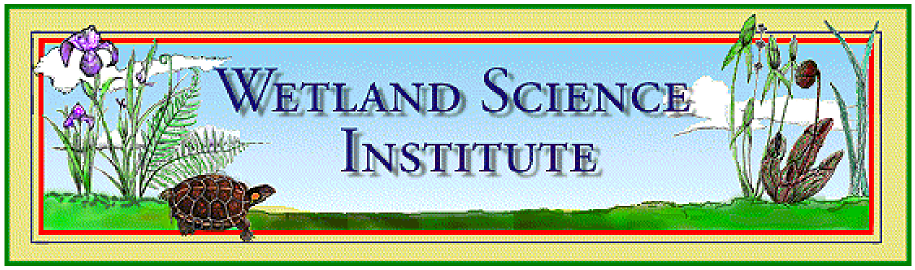
USDA United States Department of Agriculture

NRCS Natural Resources Conservation Service

Wetland Science Institute



Wetland Restoration, Enhancement, and Management

January 2003

Acknowledgments

Wetland Restoration, Enhancement, and Management may be identified as a United States Department of Agriculture (USDA), Natural Resources Conservation Service (NRCS) publication; however, it is the product of many individuals from many government entities, wildlife organizations, and academic institutions. In all cases the identity of the authors and their affiliate organization are preserved on each paper's entry, and my sincere appreciation is conveyed to all contributors. I specifically thank the following individuals who made significant contributions to this publication either through the direct offering of papers for inclusion or by working within their organization to stimulate the production of papers by others:

Phil Covington, Ducks Unlimited, North Little Rock, Arkansas

Randall Gray, NRCS, Hillsboro, New Mexico

Chris Hoag, NRCS Plant Materials Center, Aberdeen, Idaho

Mary Mattinson, NRCS, National Cartography and Geospatial Center, Fort Worth, Texas

Marcus Tidwell, NRCS, Hazen, Arkansas

Paul Rodrigue, NRCS, Wetland Science Institute, Oxford, Mississippi

Michael Whited, NRCS, Wetland Science Institute, Hadley, Massachusetts

Norman C. Melvin, III, Ph.D.
Wetland Science Institute
USDA-NRCS
Harrisburg, Pennsylvania

To Contents

Introduction

Wetlands are defined as "lands that have a predominance of hydric soil; are inundated or saturated by surface water or ground water at a frequency and duration sufficient to support a prevalence of hydrophytic vegetation typically adapted for life in saturated soil conditions; and under normal circumstances do support a prevalence of hydrophytic vegetation" (1985 National Food Security Act, as amended). Historically, wetlands in the United States are synonymous with loss. The U.S. Fish and Wildlife Service (USFWS) National Wetlands Inventory *Status and Trends Report* (1991) estimates that the original wetland acreage in the coterminous United States at the time of European settlement was 221 million acres. This same report estimates that 103 million acres (or 47%) remain, and losses continue on an annual basis. However, recently the rate of wetland loss has slowed partly the result of changes in public opinion towards the value of wetlands and also by Federal and State wetland legislation intended to conserve the resource.

At the same time wetland conversions have slowed, efforts to return wetland conditions to drained and degraded wetlands have increased. Wetland restoration and enhancement efforts by the United States Department of Agriculture, Natural Resources Conservation Service (Wetland Reserve Program), U.S. Fish and Wildlife Service (Partners for Fish and Wildlife Program), Ducks Unlimited, The Nature Conservancy, and other public and private organizations are impacting the recovery, conservation, and preservation of wetland resources throughout the United States. As of November 2001, the Wetland Reserve Program had enrolled 1,074,245 acres of converted and degraded wetland into the program since its inception in 1992 and the USFWS has reestablished 464,816 acres of wetlands through their Partners for Fish and Wildlife Program.

Wetland Restoration, Enhancement, and Management is designed to assist the NRCS field level of operation in their work by providing the most recent technical information available on specific topics. The publication is a compilation of papers on specific issues written by experts in that field. Each paper is an individual submission and stands alone, connected to the other papers in the publication only by topic similarity. In this way, individual papers can be updated and new papers added as wetland technology evolves. The topic papers are grouped into four sections, each a phase in the restoration process.

- Section I is information on techniques used to restore and enhance vegetation, hydrology, and wildlife benefit.
- Section II focuses on monitoring.
- Section III consists of papers related to management of specific species of wildlife, vegetation (beneficial and noxious), and habitats.
- Section IV is papers on restoration and enhancement techniques important from a regional perspective.

Papers listed in the Contents with **bold** type are complete and included in this issue of the technical note. Those papers in faint type are planned for future submission. This complete publication with all subsequent additions is available to be copied from the NRCS Wetland Science Institute Web site (<www.pwrc.usgs.gov/wli/default.htm>).

This publication stems from "lessons learned" over time by restorationist, foresters, wildlife biologists, botanists, engineers, and practitioners of wetland management. The papers included are our most up-to-date knowledge on the topic and information on practice application; yet, it is only a snapshot in time. Time will, in turn, further improve our understanding of wetland ecosystems. This knowledge will lead to a refinement of restoration techniques and the development of new ones. And these new techniques will affect future success. It is intended that as the science of restoration, enhancement, and management evolves, so will the *Wetlands Restoration*, *Enhancement*, *and Management* publication.

Norman C. Melvin, III, Ph.D.
Wetland Science Institute
USDA-NRCS
Harrisburg, Pennsylvania

Wetlands Restoration, Enhancement, and Management

Cover

Contents

United States
Department of
Agriculture

**Natural
Resources
Conservation
Service**

Wetland Restoration, Enhancement, and Management

Section I

Wetland Restoration and Enhancement Techniques

I.C.1 Restoring hydrology —Structures

(Paul Rodrigue, NRCS Wetland Science Institute, Oxford, Mississippi, December 2001)

Purpose

This paper provides guidance on the types and use of various water control structures in wetland restoration or enhancement projects. It provides an overview of application to a wetland situation as opposed to a production agriculture environment.

Contents

Structures

Wetland hydrology restoration often involves the use of various types of structures to restore, enhance, or regulate hydrology on the restoration site. Structures typically used in wetland restorations include dikes, low berms, diversions, grade stabilization structures, water control structures, excavated and embankment ponds, and de-leveling features, such as depressions, wildlife islands, swales, and sloughs.

Structure design requirements should be developed based upon the wetland application rather than traditional production agriculture requirements. For example, the longer detention times and storage available in wetland restoration areas should be considered in the design of outlet structures.

Structural features should represent the natural form found in the surrounding landscape. Features, such as dikes, should be set back from property lines and roads to prevent duplication of their linear form. Features should be nonlinear and random as found in nature. Following a natural contour line is preferred to following a property line.

Ditch plugs

In some areas hydrology has been removed by a surface drainage system. Ditch plugs (often a component of Wetland Restoration (Conservation Practice Code 657)) are used to reduce the effectiveness of this system by partially filling the ditch at selected points. This should be done to ditches that drain only the restored area.

Law in most states requires that runoff from and to adjacent land owned by different landowners maintains the traditional inflow and outflow points, and flow rates. Therefore, ditches that drain these properties should not be modified, and lateral measures should be applied instead.

Rice levees (low berms)

A simple method to prevent runoff and increase onsite hydrology is to install rice levees (low, narrow berms) through the restored area, especially if a Bottomland Hardwood forest restoration. Small levees are established on contour through the area and installed on a set contour interval (for example, 0.2-foot vertical spacing). Rice gates or other simple devices can be installed to provide fixed overfall points to prevent the levee from being cut by runoff flows, thereby improving longevity. These rice levees provide a temporary series of impoundments to increase hydrology until nature (debris, vegetation, and wildlife) establishes its own hydrology onsite. Rice levees should be considered a temporary measure although remnants of these levees may last for decades. This practice does not interfere with the runoff from adjacent properties.

Dikes

Wetland restoration projects often involve the use of dikes or berms to contain water for wildlife benefits. The dike standard in a state should be updated to recognize the application to a wetland restoration system (low storage depths, which would have minimal negative impacts if breached). Top widths, freeboard, and side slopes are the critical aspects in wetland restoration esthetics and wildlife needs.

While rice levees are considered temporary, dikes (Conservation Practice Code 356) can be built as permanent structures with low maintenance requirements. Dikes can be built around the perimeter of an area to retain runoff on it. They can also be built across swales to form low dams in them. If they are built parallel to drainage features, they can prevent inflow from the wetland site to the ditches, but allow the ditch to remain open for drainage of adjacent land. The top width can be set to accommodate the required vehicle traffic for access and maintenance.

Dikes should be 3 feet or less in height for wetland conditions. Multiple dikes may be required to flood large areas with typical, shallow wetland depths. A series of smaller, shallower wetlands created by a series of low dikes may be preferred to a single, large, deepwater pond and embankment, such as one built by the pond embankment standard (Conservation Practice Code 378).

To protect the dikes from washout, spillways should be installed in the dikes to handle anticipated runoff and flow rates without causing erosion of the dike. The type of spillway is dictated by the runoff design flows and the desire to manage water elevations. Repair or replacement cost may be lower than constructing for a large event.

Impoundments created by dikes should be checked to verify that they do not extend off the site.

Often a dike-like structure is used; usually called a berm (some people call them rollover dikes, implying that floodwater would overtop them). Overtopping is the critical time. High water always finds the lowest point, and that is where erosion of the berm is most likely to begin. For level berms, establish a control point, such as a vegetated or reinforced chute or spillway, to protect the dike from washing out. Otherwise, where they will overtop is unknown. Low, flat berms (only a foot or two high) with about 20:1 slopes can be oriented along the contour. An engineer (using some artistic license) can adapt existing practice specifications to wetland restorations where loss of life and property are not involved.

Water control structures

Water control structures are used in wetland restoration to help establish and manage hydroperiod by managing water surface elevation upstream of the structure. These structures may be designed with or without drawdown capability (inlet invert above wetland bottom). Fixed crest spillways are preferential to reduce or eliminate operation and management requirements or errors (unintended drawdown, delay in establishing inundation).

An operation plan should be in place for any water control structures along with any required compatible use permits.

Typical water control structures

Stoplog structures

A stoplog structure may be the familiar flashboard riser attached to a circular conduit through a dike, or it may be a straight, weir-type, open flow structure. The removal or insertion of stoplogs controls the water level. The stoplogs can be wood, metal, or plastic. Metal stoplogs may be preferred to eliminate beaver damage, floatation of stoplogs, and shrink/swell problems.

Outlet rates should take into account the storage capacity of the wetland. The wetland storage attenuates the peak flows. One method of conducting such an analysis is to use the quick flood routing procedure in exhibit 11–4, chapter 11, Engineering Field Handbook.

Flashboard riser

Sizing for row crop production is different than for wetland restoration where removal times are longer (no removal requirement to prevent crop damage) and detention capacity (natural wetland function) is greater (fig. I.C.1–1).

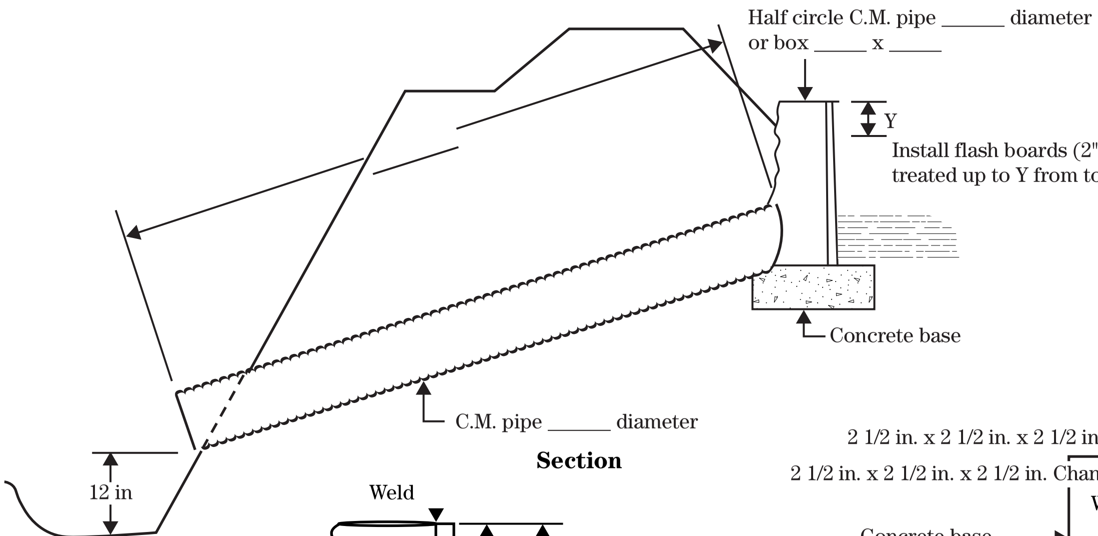
Chutes

Chutes can be installed over a dike, low berm, or embankment and act as a fixed crest spillway, where no drawdown is required (permanent water). Chutes can be vegetated spillways, or reinforced with permanent turf reinforcement material, concrete, riprap with geotextile, in-filled cellular confinement material with geotextile, gabions, or concrete block with geotextile. These chutes act as broad-crested weirs (fig. I.C.1–2).

Open drop/weir

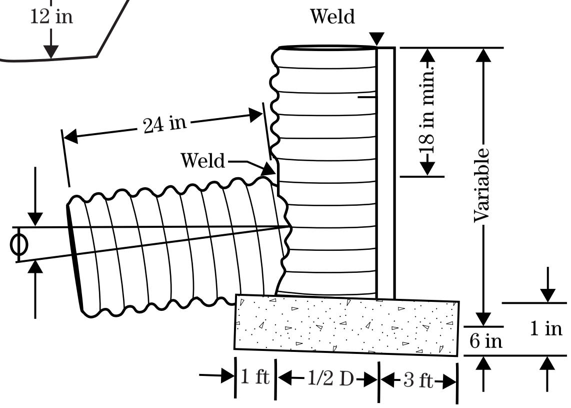
Open flow straight drops function as weirs (fig. I.C.1–3). Prefabricated metal (aluminum, steel), concrete, gabions, and other such material may be used for construction.

Figure I.C.1–1 Flashboard riser

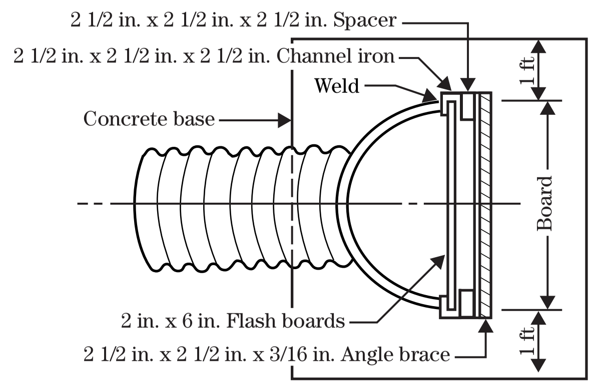


Section

Estimated Quantities		
Item	**Unit**	**Amount**
Earth fill	Cubic yard	
Excavation	Cubic yard	
C.M. pipe		
Diameter	Linear foot	
Diameter	Linear foot	
Concrete	Cubic yard	
Flashboard	Linear foot	
——x——	Each	

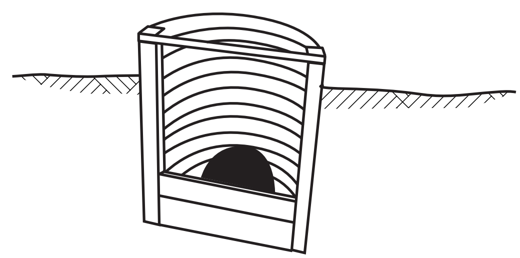


Elevation



Plan

Pipe (in)	Riser (in)	Box length x width (in)	Y (ft)	Concrete (cu yds)
12	18	9x18	.6	.62
15	24	12x24	.7	.74
18	30	15x30	.8	.88
21	36	18x36	.9	1.02
24	42	21x42	1.0	1.17
30	54	27x54	1.3	1.51
36	66	33x66	1.5	1.88



Isometric

Figure I.C.1–2 Chute installed over a dike acts as broad-crested weir

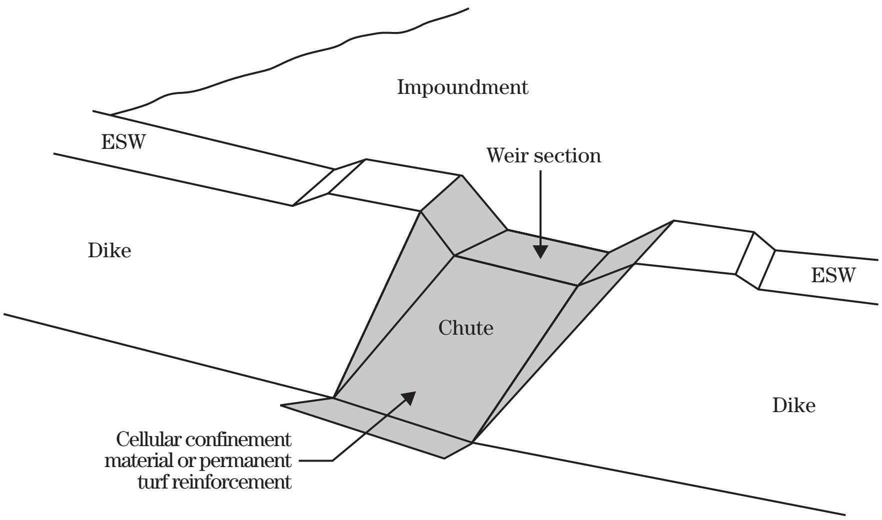


Figure I.C.1–3 Open flow straight drops (photo courtesy of Bobby Massey, Ducks Unlimited)

Additional material

NRCS Engineering Field Handbook:

- Chapter 6, Structures
- Chapter 11, Ponds and Reservoirs (quick-flood routing methodology, exhibit 11–4)
- Chapter 13, Wetland Restoration, Enhancement, or Creation

Ducks Unlimited Wetlands Engineering Manual:

http://www.sedlab.olemiss.edu/projects/rodrigue/papers.html

Standard drawings for open flow structures:

http://www.wcc.nrcs.usda.gov/wtec/wtec.html

http://www.oh.nrcs.usda.gov/engineering/cadd2.htm

I.C.2 Restoring hydrology—Microtopography and macrotopography

(Paul Rodrigue, NRCS Wetland Science Institute, Oxford, Mississippi, December 2001)

Purpose

This paper provides guidance on the establishment of micro/macrotopography as part of a wetland restoration or enhancement project. It also provides guidance on the evaluation of hydroperiod to ensure a successful hydrology restoration. Examples of micro/macrotopographic features are provided along with the Indiana Biology Technical Note No. 1, Using Micro and Macrotopography in Wetland Restoration (courtesy of Dave Stratman, biologist, USDA, NRCS, Indianapolis, Indiana).

Contents

Definitions

Microtopography—Topographic features with a vertical relief of less than 6 inches. Includes small depressions, swales, wallows, and scours that would hold water for a short (hours to days) time after a rainfall, runoff, or flooding event. Small ridges that are rarely inundated are included here as well.

Macrotopography—Topographic features with a vertical relief of 6 inches to several feet. Includes deeper depressions, swales, and sloughs that hold water for a significant (weeks to months) time after a rainfall, runoff, or flooding event.

Hydroperiod—The timing, depth, and duration of saturation and inundation. Hydroperiod should be considered over a long-term climatic scenario (10 years minimum) rather than using one *normal* or *typical* year that can provide misleading results.

Restoring hydrology

Restoring hydrology on a wetland restoration requires the establishment of microtopography and macrotopography in the wetland landscape. The amount, degree, and type of topography to be developed are based upon the purpose and objectives of the wetland restoration.

Once plant and wildlife needs are addressed, an appropriate hydroperiod (or hydroperiods) can be planned. A diversity of hydroperiods, achieved with a diversity of topographic features, can provide the wetland characteristics required in the wetland restoration plan. Hydroperiods should be planned for the appropriate plant (woody and herbaceous) and animal species (waterfowl, amphibians, insects) expected to inhabit the diverse restoration site.

By providing micro- and macrotopography, a diversity of hydroperiods will exist on a site, from permanent water to short-term, seasonal ponding.

Examples

1. To promote amphibian habitat, areas of nonpermanent water should be developed to prevent predation by fish. Therefore, inundation should be planned as temporary or seasonal. Microtopographic depressions and swales may best fit these requirements.

2. To promote wading bird habitat, areas of receding water that continually expose new shoreline are desirable. Therefore, inundation should be planned as long-term seasonal or semi-permanent with a large area-to-depth ratio (flat side slopes, 10:1 or flatter).

3. To promote waterfowl habitat, areas of open water for resting, feeding, or brood-rearing are always desirable on at least part of the site. Therefore, inundation should be planned as permanent. Deep ponds or swales should be planned with steeper side slopes (5:1 or steeper) to reduce evaporative surface area.

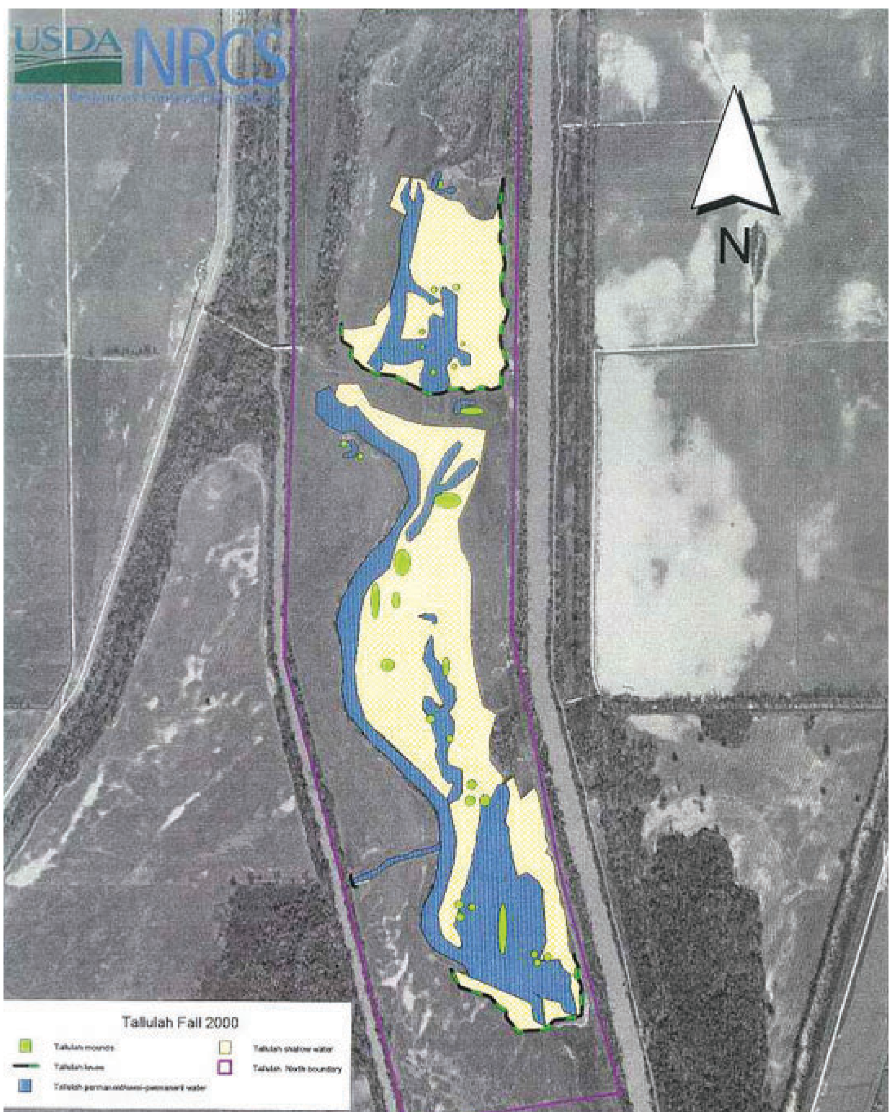
Limited micro/macrotopography

In some circumstances, especially in bottomland hardwood restoration (BLH), sites may be afforested without any major hydrologic manipulation if a sufficient quantity and diversity (e.g., isolated depressions) of hydrology are present on site. If sufficient microtopography is not present (area has been smoothed or leveled), restoration of depressional features must be included in the restoration plan. In some cases the areas are already subject to out-of-bank flooding by rivers and streams (these areas are sometimes classified as Farmed Wetlands). In these areas small berms can be made to hold water for a longer time after the floodwater recedes. In other cases the drainage is poor so that sufficient periods of soil saturation occur, particularly during winter and early in spring. Depressions created in these areas will provide some open water.

Planning micro/macrotopographic features

Constructed micro/macrotopographic features should emulate the natural form of the surrounding landscape. Features, such as dikes, should be set back from property lines and roads to prevent duplication of their linear form (fig. I.C.2–1). Features should be nonlinear and random as found in nature. Following a natural contour line is preferred to following a property line.

Figure I.C.2–1 Example of serpentine dikes being set back from linear property lines (Louisiana NRCS)



Stream analogy

Natural streams have certain features: meanders, riffles, glides, runs, pools, thalwegs, bars, headcuts, and natural levees. Wetland hydrology restoration should consider synonymous features depending upon the wetland type (table I.C.2–1).

Random and nonlinear (e.g., serpentine rather than straight dike) planning result in a landscape that represents the nonuniform and irregular landscape patterns found in nature. In his report on the Lower Mississippi Valley, Saucier (1994) states:

> *Geomorphic processes determine how, when, where, what, and if sediments are deposited. Landscapes, however, involve the products of geomorphic processes in the context of regional settings and geologic controls. Scale and time become important factors. Meander belts, for example, consist of numerous individual natural levees, abandoned channels, and other landforms, but the types, number, and distribution of meander belts depend on valley size, shape, slope, interactions among meander belts, and time.*

Table I.C.2–1 Hydrology features

Stream	Wetland
Pool	Depression or dugout, temporary to permanent
Run	Flow path, short duration
Glide	Shallow flow, mid-duration
Bars	Islands, uplands
Meanders	Serpentine sloughs and serpentine levees
Headcuts	Control structures - drawdown capability, fixed crest - no drawdown
Natural levees	Dikes, diversions
Riffles	Debris dams, check dams
Thalwegs	Stable concentrated flow areas

Micro/macrotopography planning should aim to look as if there is no planning at all. Flow patterns should snake through the area rather than show a rigid drainage pattern, and only inflow and outflow points should be maintained to respect the rights of adjacent landowners. Flow patterns through sloughs should have areas that equate to the runs, glides, pools, and riffles of flowing streams.

Hydrology considerations

Water source—The source of water is fundamental for evaluating a site's potential wetland hydrology restoration. Sources of water may include direct precipitation, runoff from contributing drainage area, groundwater discharge, or riverine or lake flooding. Pumping, from groundwater or surface water, may also be a water source for the site, but a source that has a significant operation and maintenance component. Pumping can provide early season water or water in dry years.

Direct precipitation and runoff are easily evaluated from available weather data. Riverine or lake flooding can only be easily evaluated if stage data are available. Inputs from groundwater discharge are difficult to assess and evaluate without extensive monitoring.

Storage capacity—The amount of water the topographic feature can hold, and the stage:storage:area relationships (saucer shaped as opposed to bowl shape) are important in establishing the timing and duration of inundation and changes that occur seasonally. The stage:storage:area relationships relate to the shape of the feature, how the surface area will change as water is added (rainfall, runoff) or lost (evaporation, plant transpiration, deep seepage). A saucer-shaped feature has large changes in surface area in response to small changes in water losses or additions. Conversely, a bowl-shaped feature has small changes in surface area in response to large changes in water losses or additions.

Saucer-shaped features tend to be temporary and seasonal with large areas exposed as water is lost (wading bird habitat). Bowl shaped features tend to be semi-permanent to permanent with smaller exposed areas as water is lost, but providing long-term water to support bird species and amphibians.

This storage effect also affects temporary storage and the requirements for outflow devices (see exhibit 11–4, chapter 11, Engineering Field Handbook).

Water losses—Water losses in wetland topographic features include deep percolation, evaporation from wet surfaces, and transpiration through vegetation including adjacent trees that have roots using water from the wetland feature. Outflow is also a water loss.

Operation and maintenance

The hydrology restoration plan should minimize future operation and maintenance requirements as the site matures. The site should have no excessive erosion or sedimentation problems that would fill microtopographic features at higher than normal rates.

Hydroperiod management can be minimized by having fixed crest outlet elevations. This is preferred. However, hydroperiod can be manipulated by the use and management of water control structures, such as stoplogs, gate valves, adjustable riser pipes, or flap gates (see section I.C.1), Restoring hydrology—Structure, in this technical note.) If this method is used, determine if a compatible use permit is required.

An overall management plan must be developed for hydroperiod manipulation including the operation of control structures.

Creative borrowing

Borrow areas for dikes or embankments can be planned as permanent pools or deepwater habitats. Excess material can be used to create islands in water features or upland areas in flatlands.

Where possible, excavation as a source of fill for dikes should be away from the dike. This prevents the establishment of permanent water against the dike and the inherent possibility of providing habitat for burrowing rodents.

Evaluating hydroperiod

Areas receiving natural flooding from lakes and rivers may not need evaluation beyond determining frequency of flooding (gage data). However, depressional features dependent upon rainfall (runoff) for inundation need to be evaluated.

Hydroperiod should be evaluated in some fashion to determine if the desired hydrology restoration goals will be achieved. Evaluation can be quite simple or very elaborate depending upon the accuracy and detail required.

Hydroperiod is determined based upon storage, rainfall, evapotranspiration (ET), and drainage area. Make pool deeper to offset lack of drainage area if permanent water for waterfowl is desired.

SPAW model (ftp://c100.bsyse.wsu.edu/pub/spaw/)—SPAW (Soil-Plant-Air-Water) is a water budget model. It consists of two basic components: a field component and a pond component. For a wetland restoration analysis, the field component would be used to evaluate runoff from a contributing drainage area. The pond component would be used to evaluate the hydroperiod (timing, depth, duration) of the wetland area. All processes are evaluated, and long-term, continuous simulations can be performed. The program is relatively easy to use, and input data are readily available.

Spreadsheet water budget—A simple water budget (rainfall, evaporation, runoff) can be set up in a spreadsheet for individuals experienced in spreadsheet use. The budget can be made simple or extremely complex depending upon the user's needs and experience level.

Example

An example result from a simple spreadsheet water budget is shown in figures I.C.2–2 to I.C.2–5. Daily values of rainfall and pan evaporation were the inputs.

As can be seen from figures I.C.2–2 to I.C.2–5, the concept of *permanent* water is relative. The planner must decide how many times in a period of years the site can go dry and still meet the planned requirement of permanent water. These figures of hydroperiod can be used to establish if the site will meet the hydrology needs of plant and animal support or control.

Figure I.C.2–2 Direct precipitation, 12 inches maximum storage (outflow above 12 inches), and no contributing drainage area. Result is many periods will be dry, water is not permanent. To make water more permanent, increase storage depth or add recharge area using diversions and other such structures.

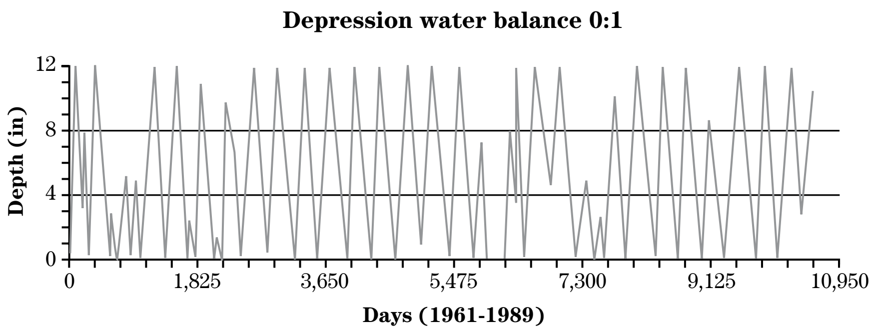


Figure I.C.2–3 Depth increased to 24 inches (outflow above 24 inches), no contributing drainage area. Result is water that is permanent in most years.

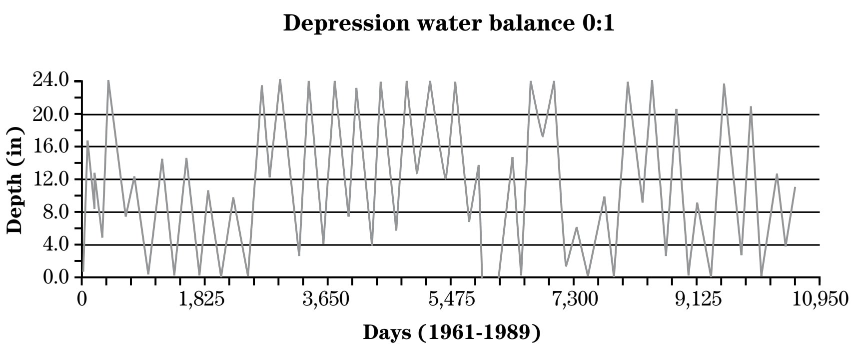


Figure I.C.2–4 Depth 24 inches, contributing drainage area equal to surface area of wetland. Result is a permanent water surface, with minimal exceptions.

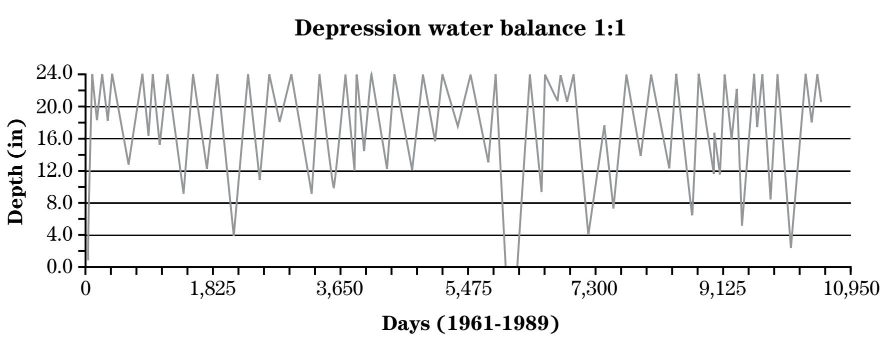
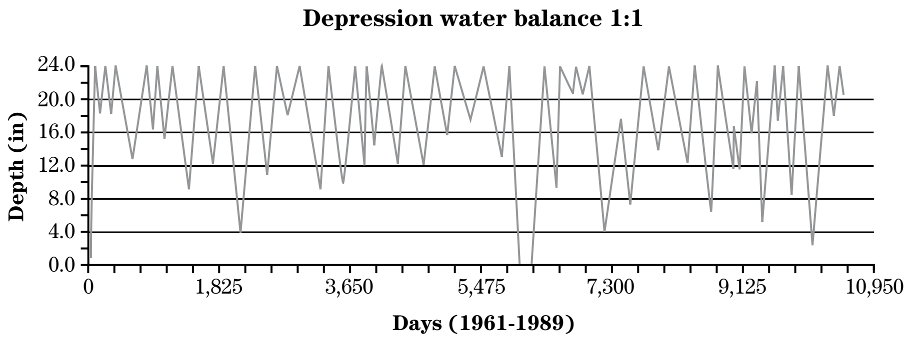


Figure I.C.2–5 Shows a site where storage depth is used to make up for contributing drainage area. The depth is increased to 48 inches (outflow above 48 inches).



Costing out micro/macrotopography

Because of the random and unique shapes and sizes desired for the creation of micro/macrotopography in the wetland restoration landscape, traditional methods of payment are cumbersome and time-consuming. To ensure that all parties are properly billed and paid for the work performed, a method of payment must be established agreeable to all parties.

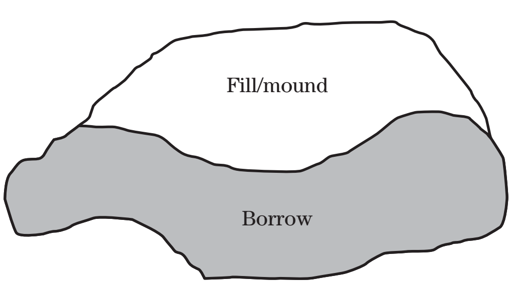
The need for exact layout and checkout, with its inherent costs, can be replaced with typical sections/features that can be laid-out and checked-out simply and quickly with minimal dimension checks, such as average length, width, and depth.

Dikes: By minimum number of X-sections/segments
Features: Per item/unit based on size/area

Examples (costs are for example purposes only)

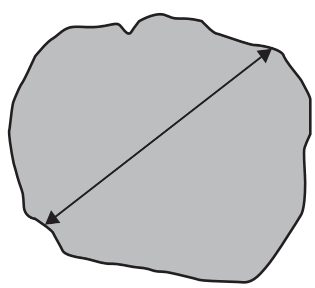
Sloughs:

1' x 30' x 100'	$275 (each)
2' x 30' x 100'	$550
2' x 30' x 1,000'	$5,500



Depression/pothole:

50' dia. 1' deep	$180 (each)
50' dia. 2' deep	$360
100' dia. 2' deep	$1,400



Additional resource materials

NRCS Engineering Field Handbook:

- Chapter 13, Wetland Restoration, Enhancement, or Creation
- Chapter 19, Hydrology Tools for Wetland Determination

Ducks Unlimited Wetlands Engineering Manual:

http://130.74.184.149/Rodrigue/papers.html

References

Saucier, Roger T. 1994. Geomorphology and quaternary geologic history of the Lower Mississippi Valley. U.S. Army Corps of Engineers, Mississippi River Commission, Vicksburg, MS.

Using Micro and Macrotopography in Wetland Restoration

Indiana Biology Technical Note No. 1

This document is intended to be used as a tool to assist in the planning of wetland restorations where the natural topography of the site has been eliminated. The planner is encouraged to be creative when developing the restoration plan. The concepts within can also be used whenever the development of macrotopographic features are desired.

WHAT IS MACROTOPOGRAPHY?

Background Undisturbed wetland systems in Indiana typically consist of complexes that contain a diversity of topographic relief from extremely shallow areas with minor ridges (microtopography) to deeper wetland habitats that include some upland characteristics (macrotopography). When wetlands are drained or altered, they normally lose most of their micro and macro topographic relief through land leveling or other agricultural activities.

Macrotopographic features are wetland "ridge and swale" complexes whose basins are depressional in landscape position and occur on terraces and in floodplains. The basin areas are normally from 0.1 acre to 5 acres in size with depths running from 0-30 inches, depending on the landscape position. These types of wetlands can be found in a multitude of shapes ranging from simple circular basins, to complex amoeba-like outlines, to meandering scours. Ridges (linear) and mounds (circular or elliptical) make up the "upland" component of macrotopographic features that normally do not exceed 30" in height. Together, the ridge and swale features form ephemeral wetlands that hold water from only a few weeks to several months during the year.

Microtopographic features are normally thought of as those shallow depressions with less than 6 inches of depth between the swales and ridges. Examples of microtopography can be seen in flat fields where shallow "sheet" water stands for short durations after a rain. Within the scope of this document, macrotopography will be assumed to include microtopographic features.

WHY IS THE DEVELOPMENT OF MACROTOPOGRAPHY IMPORTANT?

The development of macrotopographic complexity creates a diversity of water regimes (hydroperiods) which can increase water quality, provide flood storage, and enhance the development of a more diverse vegetative community. This results in greater overall wildlife benefits through the development of a variety of habitats. The dispersal, germination, and establishment of plant species, and the life cycles of many amphibians, reptiles, and other wildlife species are dependent on variations in the timing, depth, and duration of flooding.

Pickerel Frog

Food In the spring, shallow, ephemeral wetlands warm up before larger, deeper bodies of water, and provide important seasonal forage for shorebirds, waterfowl, nonmigratory bird species, and other wildlife. These types of wetlands produce significant amounts of protein-rich invertebrates including snails, worms, fairy shrimp, midge larvae, spiders, backswimmers, diving beetles, dragonflies, and damselflies. Organic (woody and herbaceous) debris, roots, leaves, and tubers from aquatic vegetation are additional food sources and provide substrates for macroinvertebrates.

Habitat Wetland restoration plans that include undulating landscape features create a diversity of habitat types. Swales, oxbows, potholes and other macrotopographic basins provide varying hydroperiods from short-term ponding to seasonal and semi-permanent water conditions. A wetland, or wetland complex, with multiple hydroperiods can support a variety of habitat zones. Scrub-shrub, submergent, emergent, and floating-leaf communities (e.g., duckweed) are examples of herbaceous aquatic habitats. A diverse wetland plant community benefits numerous species of wildlife including many fur-bearing mammals, waterfowl, shorebirds, wading birds, amphibians and reptiles. Because native plants provide the best overall habitat, are essentially self-sustaining, and tend to be non-invasive, only native vegetation should be planted. Note that Conservation Practice Standard 657, Wetland Restoration, has an extensive list of native wetland plant species.

Low-level mounds or ridges (maximum 30 inches) are considered to be a component of macrotopography, and can greatly increase the biological diversity of restoration sites when combined with basins. Amphibians, for example, tend to have small home ranges. Thus, having a diversity of wetland types in close proximity to terrestrial habitats within the project area will support the greatest populations.

PLANNING

When developing macrotopographic features, the planner should determine the target species (i.e. species of concern) and review historical aerial photography to determine the appropriate features to include in the restoration project.

Tiger Salamander

Amphibians and Reptiles A primary focus of macrotopography development is the creation of habitat for frogs, toads, salamanders, newts, turtles, and snakes. These amphibians and reptiles are known as herpetofauna or commonly called "herps". Amphibians are an especially diverse group and require wetlands with differing hydroperiods and habitat types. Because macrotopographic basins are often completely dry by summer or early fall, they are normally free of fish. Occasionally pools do retain water year round, but due to warm water conditions that create low oxygen levels, they still do not support fish populations. This is important because fish are primary predators of larval, tadpole, and adult amphibians. In general, sites flooded for longer periods will have more predators of amphibians.

The timing and duration of flooding are important factors that dictate which amphibians will use a particular wetland. Amphibian species are extremely variable in their habitat requirements. Most breeding occurs from May through August, with eggs hatching anywhere from 4 to 20 days later. Complete metamorphosis may take an additional 7 weeks to 3 months. Some species may need as much as a year to develop, with a few species even over-wintering as tadpoles, requiring permanent water. Table 1 (modified from Knutson et. al.) is an example of the diversity in preferred breeding periods and guild associations, for a study in an Iowa and Wisconsin.

Table 1[1]

			Breeding[2]		Nonbreeding[3]			Hibernation[4]		
Common name	Scientific name	Breeding period	Perm. water	Temp. water	Water	Forest/ litter	Open	Water	Forest/ litter	Ground
Wood frog	Rana sylvatica	Mar.-Apr.	N	Y	N	Y	N	N	Y	N
Chorus frog	Pseudacris triseriata	Mar.-May	N	Y	N	Y	Y	N	Y	N
Spring peeper	Pseudacris crucifer	Mar.-Summer	N	Y	N	Y	N	N	Y	N
N. leopard frog	Rana pipiens	Apr.-June	Y	Y	Y	N	Y	Y	N	N
Pickerel frog	Rana palustris	Apr.-mid June	Y	N	Y	Y	Y	Y	N	N
American toad	Bufo americanus	Apr.-June	Y	Y	N	Y	Y	N	Y	N
Eastern gray treefrog	Hyla versicolor	May-Aug.	Y	Y	N	Y	N	N	Y	N
Cope's gray treefrog	Hyla chrysoscelis	May-Aug.	Y	Y	N	Y	Y	N	Y	N
Cricket frog	Acris crepitans	May	Y	N	Y	N	N	N	Y	N
Green frog	Rana clamitans	Mid May-July	Y	N	Y	N	N	Y	N	N
Bullfrog	Rana catesbeiana	May-July	Y	N	Y	N	N	Y	N	N
Fowler's toad	Bufo woodhousii	Mar.-Aug.	N	Y	N	N	Y	N	N	Y

[1] Species that can successfully survive or reproduce in a habitat during the identified life-history phase are identified with a Y; those that do not with an N.
[2] Will breed in permanent water or temporary (ephemeral) ponds.
[3] Active, nonbreeding portion of the year is spent in the water or along the water edges, in trees or forest litter, or in open, nonforested habitats (grasslands).
[4] Hibernation or estivation period is spent in or near water, in forest litter, or underground.

In Indiana, the species that metamorphose their life cycle by early summer are the ones we need to target. Therefore, **macrotopographic basins should be designed to keep water available until at least mid-July.** Note that the process of a wetland drying out is beneficial. It eliminates insect and vertebrate predators, allows seeds to germinate, and exposes detritus to processes of oxidation thereby releasing nutrients.

When planning a site for amphibian and reptile habitat, macrotopographic features should make up approximately 30-50% of the area. The water (swale, meander, etc.) and the upland habitat (mound) acreage are combined to get the percent of macrotopographic features. It can be assumed that for every acre of water created, an additional acre of mound is created. **Table 2** can be used to record the planned macrotopographic features.

Table 2

Field Number	Field Size (acres)	Basin Number	Basin Amount (acres)	Macrotopography Description	Associated Habitat Mounds (height(#))

Where restoration sites have a designed water level, such as those with levees and control structures, approximatey 30% of the area should have macrotopographic features. Consider concentrating macrotopographic features in and near the more shallow water reaches.

Where restoration sites do NOT have a designed water level, such as in floodplains where high stream flows would destroy levees and control structures, approximatey 50% of the area should have macrotopographic features. Note that in these landscapes, the macrotopographic basins may provide the only standing water on the restoration site. Consider concentrating the deeper macrotopographic features in the lower elevations of the site, and shallower features in the higher elevations.

Shorebirds Shallow, ephemeral wetlands provide an abundance of aquatic invertebrates that are a critical food source for shorebirds during migration. Most shorebird species will utilize wetland habitats with water depths from 0-3 inches, and will rarely forage in water depths greater than 6 inches. Maximizing areas which provide conditions from mudflats through 3 inches deep during spring and late summer will provide the greatest benefits for migratory shorebirds.

Waterfowl These same shallow basins provide important invertebrate forage for waterfowl, particularly during spring migration when nutrient needs prior to nesting are high. In addition, several species of dabbling ducks (e.g. mallards and blue-winged teal) will utilize temporary wetlands for pair bonding and mating. Although these temporary ponds may not have water long enough to provide brood habitat in most years, they serve an important function in distributing pairs across the landscape and allowing for courtship rituals. Visually isolating basins, or portions of basins, through irregular shaping will particularly benefit species such as mallards which are more territorial. When combined with semi-permanent basins in close proximity, macrotopographic basins contribute to excellent wetland complexes for water fowl breeding.

Soils It is important for the planner to identify those portions of the restoration site which have hydric soils or soils that will most likely respond to macrotopographic development. Look for soils that have low permeability, a restrictive under-lying layer, or high water tables.

Sites which have soils that are hydric due only to flooding may not be appropriate if the soils are well drained and are not very frequently flooded. In these cases, it may not justify the expense of creating macrotopography and the planner should consider only vegetative restoration measures. If it is unclear whether or not there is sufficient hydrology to maintain the needed water levels within the basin areas, a water budget should be calculated.

Succession and Long-term Management Succession of wetlands is a natural process that can result in significant habitat changes over time. Primary changes include, for example, the development of aquatic macrophytes, invasion of wetlands by trees and shrubs, and canopy closure over wetlands embedded in forested landscapes. Such changes can alter the species composition of wetlands over time by selecting for species that favor or can tolerate later successional stages. Early successional species will consequently be lost, thereby lowering diversity, and can only be restored by periodically reversing succession. Plans to periodically (e.g. every 10-20 years) reverse the effects of succession in some portion of all wetlands (e.g. 5-10% of the total number per year) are important to consider. Natural processes that can reverse succession vary among regions and should mimic local regimes but may include flooding, drying, and burning. Human disturbance regimes such as mowing, timber harvest, draw-downs, or even herbicides may be considered, but only with extreme caution because of possible negative indirect effects.

MACROTOPOGRAHIC BASINS

The macrotopograhic basins are described in abbreviated format as: shape/size/depth.
Where:

1) the shape is described below
2) the size is in acres
3) the depth is in feet

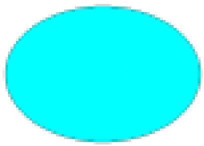
For example, a macrotopograhic basin described as Oxbow/1.5/0.5-1.0-2.0:

1) has shape #2 below,
2) is 1.5 acres in size, and
3) is composed of 3 depths (0.5', 1.0' and 2.0')

BASIN SHAPE DESCRIPTIONS

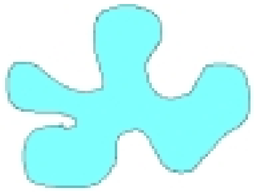
Basins should be irregular in shape. Irregular shapes increase edge and provide additional cover for waterfowl and other wildlife utilizing the site.

1) Shape: **Oval**
Description: Generally circular



2) Shape: **Oxbow**
Description: Kidney shaped with 2 lobes

3) Shape: **Amoeba**
Description: Multiple lobes with random shape, high perimeter to surface area ratio



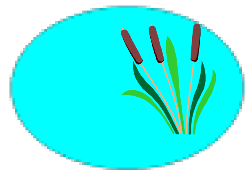
4) Shape: **Meander**
Description: Mimics an abandoned stream channel meander

DEPTH DESCRIPTIONS

AERIAL VIEW | **CROSS SECTION**

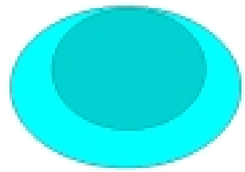
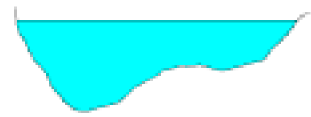
When <u>1 depth is indicated</u>:

- the basin is primarily 1 depth




When <u>2 depths are indicated</u>:

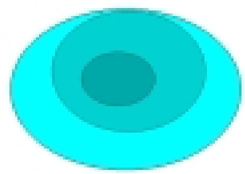
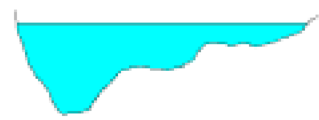
- each depth composes approximately 50% of the area

When <u>3 depths are indicated:</u>

the depths compose approximately:

- deepest depth = 20% of the area
- middle depth = 30% of the area
- shallowest depth = 50% of the area

HABITAT MOUNDS

Fill excavated from the macro-topograhic basins can be used to create multiple upland habitat conditions based on the height, shape, and location of habitat mounds. Variations in habitat mound design can provide escape areas, denning sites, nesting opportunities, and plant diversity, as well as providing visual breaks within the wetland complex. All side slopes for mounds should have a minimum slope of 6:1, but should be as flat as is feasible. **Note:** In situations where geese are a nuisance, at least 30 feet should exist between the habitat mound and any water surface. This area should then be planted with a vegetative barrier such as warm season grasses, trees or shrubs.

Where restoration sites have a designed water level, habitat mounds should vary in elevation from above to below the expected normal waterline. Approxmately 1/3 of the mounds should be 6 inches to 1.0 foot **below** the normal water elevation, 1/3 should be 6 inches to 1.0 foot **above**, and 1/3 should be **at** the normal water elevation.

Where restoration sites *do not* have a designed water level, habitat mounds primarily provide upland habitat and tend to direct water flow during flood conditions. Approximately 50% of the mounds should be 6 inches to 1.0 foot above average ground level, and 50% should be 1.0 to 2.0 foot above the normal ground elevation. Mounds should mimic the natural landscape as much as possible. For example, if the site is located on the interior of a river oxbow, ridge and swale design may be appropriate (see figures 2 and 3). When possible, place mounds in such a way as to increase meander distance by directing water flow in a path that meanders across the unit.

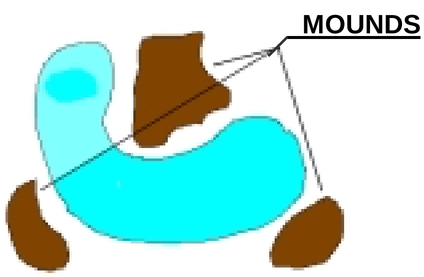


Figure 1

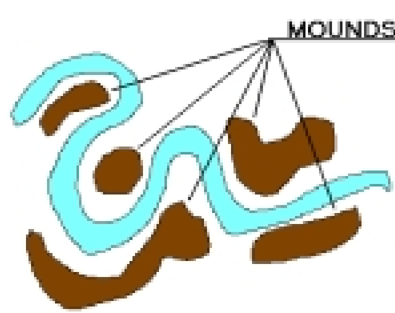


Figure 2

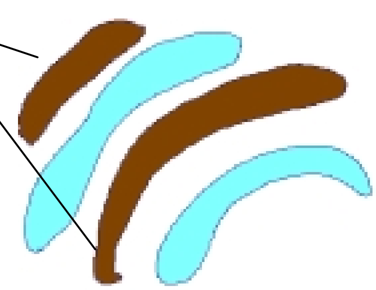


Figure 3

ADDITIONAL MODIFICATIONS

Ditches of varying depths and widths can connect basins to diversify a site. They provide additional cover for waterfowl as well as escape routes away from predators. Connection ditches may have 3:1 (or flatter) side slopes. In some cases, they can also be used for boat access to the site for hunting and recreational viewing, or to limit vehicular traffic of the site. See Figure 4.

Note: In situations where amphibians are the primary species of concern, connecting ditches should be limited because they provide access routes for predatory fish, particularly if connected to deeper, more permanent pools.

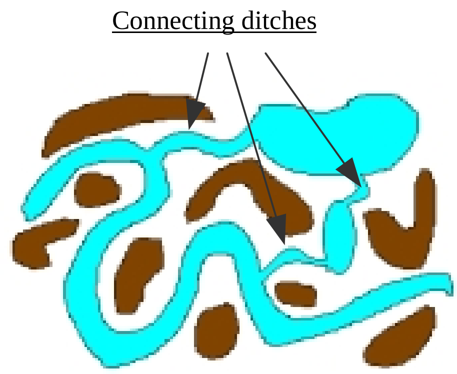


Figure 4

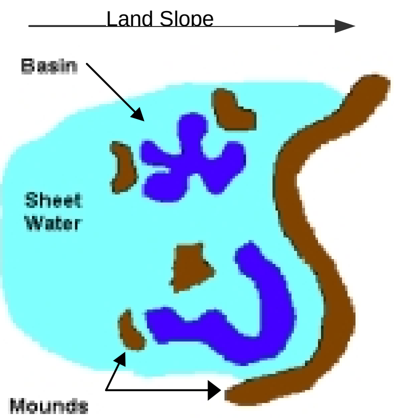


Figure 5

On gently sloping sites, an efficient means of providing shallow, "sheet" water habitat is through the creation of linear habitat mounds. The excavated material from a macrotopographic basin is used to form a low, meandering ridge on the down slope side of the basin(s). Typical heights for the mound range from 1 to 2 feet. By using the spoil in a creative manner, the total shallow water on a project site can be substancially increased. The impounded sheet water provides seasonal or ephemeral water for shallow feeders such as shorebirds, while the excavated basins provide longer hydroperiod wetland habitats. This method can also be utilized where wetland meadow conditions are desired.

CONSTRUCTION

Creative Borrowing Borrow areas for dikes or embankments can be incorporated into the development of macrotopographic features. Potholes, swales, meanders, and other shallow water habitats can serve as borrow areas for needed fill. All side slopes for basins should have a minimum slope of 6:1. Note that, when feasible, slopes should be as flat as possible. Slopes exceeding 20:1 are not considered excessive for habitat purposes. Examples of this include situations where equipment operators randomly fill their scrapers leaving shallow, single-trip borrow sites. Note that the borrow areas will result in the basins being the deepest portions of the wetland complex. In seasonal or ephemeral wetlands these areas provide a diversity of hydroperiods by holding water later into the year than the remainder of the wetland.

Rough-finish Grading The desired macrotopographic features will have rough surfaces on all side slopes and top, an undulating bottom, and a ragged shoreline.

Woody Debris

- Provides sunning and resting areas for herptiles
- Provides loafing sites for waterfowl
- Is a source for organic soil material
- Provides additional vertical and horizontal habitat
- Is an excellent substrate for invertebrates

Depending on water velocities the debris may or may not have to be partially buried. Use as needed.

ASSOCIATED TECHNICAL STANDARDS

This technical note can be used in association with the following technical standards:

- 657 Wetland Restoration
- 658 Wetland Creation
- 659 Wetland Enhancement
- 644 Wetland Wildlife Habitat Management

REFERENCES

KIEFER, J. L. U.S.D.I. Fish and Wildlife Service. 2000. *Personal Communications*, Ecological Services Field Office, Bloomington Indiana.

KING, S. L. and LICHTENBERG, J. S. *Habitat Restoration and Amphibians*. USGS National Wetlands Research Center.

KINGSBURY, B. A. 2000. *Personal Communications*, Indiana University Purdue University-Fort Wayne.

KNUTSON, M. G., SAUER, J.R., OLSEN, D. A., MOSSMAN, M. J., HEMESATH, L. M., AND LANNOO, M. J. 1999. *Effects of Landscape Composition and Wetland Fragmentation on Frog and Toad Abundance and Species Richness in Iowa and Wisconsin.* Conservation Biology 13 (6), 1437-1446.

KNUTSON, M. G., SAUER, J.R., OLSEN, D. A., MOSSMAN, M. J., HEMESATH, L. M., AND LANNOO, M. J. 2000. *Landscape Associations of Frogs and Toad Species in Iowa and Wisconsin, USA*. Journal of the Iowa Academy of Science 107: in press.

KOLOZSVARY, M. B., AND SWIHART, R. K. 1999. *Habitat Fragmentation And The Distribution Of Amphibians: Patch And Landscape Correlates In Farmland.* Can. J. Zool.:1288-1299.

LAWRENCE REGIONAL RIPARIAN TECHNICAL TEAM. 1997. *Draft Interim HGM Model For Kansas Wooded Riverine Wetlands*. Lawrence, Kansas.

SEMLITSCH, R. D., AND J. R. BODIE. 1998. *Are Small, Isolated Wetlands Expendable?* Conservation Biology 12:1129-1133.

SEMLITSCH, R. D. 1998. *Biological Delineation of Terrestrial Buffer Zones for Pond-Breeding Salamanders*. Conservation Biology 12:1113-1119.

SEMLITSCH, R. D. 2000. *Principles For Management of Aquatic-Breeding Amphibians.* Journal of Wildlife Management 64(3):615-631.

U.S.D.A. Natural Resources Conservation Service. 2000. *Techniques for Restoring Wetland Topography* (Video), NRCS Wetlands Science Institute, NRCS Watersheds and Wetlands Division.

FOR ADDITIONAL INFORMATION

DAVE STRATMAN, Biologist
USDA Natural Resources Conservation Service
6013 Lakeside Blvd.
Indianapolis, IN 46278
Phone: (317) 290-3200 ext. 354
Email: dave.stratman@in.usda.gov

Printed October, 2000

I.D.1 Restoring vegetation by natural and artificial means: an overview of considerations

(Norman Melvin, NRCS Wetland Science Institute. Laurel, Maryland, December 2001)

Purpose

This paper provides guidance on the initial steps in determining a revegetation strategy for a potential wetland restoration or enhancement project. It has decision sequence keys for use in determining whether a site can successfully revegetate through natural recourses (natural succession) or whether some level of vegetation introduction (active revegetation) will be necessary to meet the planned functions and objectives. Basic terminology used in revegetation is defined.

Contents

Vegetation directly affects the presence of wildlife, both in their overall species richness and population densities. Vegetation provides the basis for food chain support for wildlife and determines the overall community structure. The NRCS technical standards for Wetland Restoration (657) and Wetland Enhancement (659) both identify the establishment of vegetation on site as a condition of overall project success. The presence of a minimum plant species diversity, expectations of percent vegetative cover, and a timeframe for the establishment of the diversity and cover are included as part of these standards. Generally, these three criteria (diversity, cover, and time) are used as vegetative success measures in a project.

To the restorationist, vegetation decisions drive both cost and success. The decision to apply some forms of active revegetation (planting) increases project costs. Not applying vegetation may result in failing to meet the success criteria or the establishment of targeted functions. If it can be determined that directly establishing vegetation by planting or seeding can be minimized, while at the same time being relatively sure success criteria and targeted functions will be established, then project cost can be saved or redirected. When confronted with the final decision (to plant or not to plant), it is important that the choice is made based on factors that influence the ability of vegetation to produce the desired outcome over the expected timeframe. The following information is provided to assist in that critical, final decision.

Definitions

Natural regeneration—Allowing a site to revegetate on its own through the natural process of plant succession. Plant sources colonizing the site are derived from propagules present in the soil seed bank and/or dispersed by wind, animals, water, or other natural means of delivering plant materials onto the site in forms that are capable of surviving and establishing.

Active revegetation—Establishing vegetation by physically placing seed, seedlings, cuttings, or other propagules onto a site. This includes a wide variety of activities. It ranges from (1) covering the entire site with seed or propagules of selected species, to (2) establishing only the dominant species (or species that are integral components of the community, but are unlikely to disperse onto the site) and relying on natural regeneration processes to augment the remaining species diversity, or (3) adding as few as one species to enhance a specific wetland function.

Propagule—Any of a variety of plant parts that are capable of establishing a new individual. Some common examples of propagules that are used in wetland restoration and enhancement activities include seeds, cuttings, bulbs, whips, and runners.

Seed bank—Viable seeds and/or other propagules present in the soil/sediment occurring on site, or in materials transported to a site, and are capable of establishing a new individual. Fleshy propagules (bulbs, rhizomes, runners) and seed from some species (e.g., some oaks) have a limited longevity (1 year or less). The seed of some weed species may remain viable for many years if buried in the sediment. Modifying a site's soil condition by continued cultivation or altering its hydrology depletes a seed bank in a relatively few number of years. Excavation of a site to deepen a basin will remove the seed bank.

Seed wall—The standing vegetation (usually considered woody/timber) immediately adjacent to a site. In wetland restoration methodology, it is generally considered to be the source of seeds/propagules for revegetating portions of the restoration site, assuming the species are acceptable to the project objectives and have hydrology tolerances compatible with site conditions.

Decision sequence keys

Two dichotomous keys are shown as exhibits at the end of part I.D.1–1. The first key aids in determining the degree of planting/natural regeneration needed for restorations or enhancements where woody vegetation is targeted. The second considers the establishment of herbaceous vegetation on wetlands. On sites planned to include areas of both woody and herbaceous vegetation, consult both keys. The keys are set up similar to those identifying plants or animals. They are composed of a series of couplets (paired, contrasting statements). Beginning with the two number **1**'s, read both statements and choose the one that best fits the conditions present on the restoration/enhancement site slated for action. When that choice has been made, follow the **go to** directions. That will result in either a number or a letter. If the result is a number, go to the couplet prefaced with that number and repeat the process of reading the couplet and deciding which of the two best-fit site conditions. The end result of keying will be a letter (A, B, or C) that corresponds to a recommendation on how to approach revegetation. Based upon the restoration site conditions, the recommendations will be to rely on natural colonization as a means of revegetation on (A) the entire site, (B) none of the site, or (C) portions of the site.

Decision sequence keys—recommendations

The recommendation that was derived from working through these keys is the first step towards the planning of revegetation strategies to meet the project's goals and success criteria. The recommendation is based on the site's condition, taking into account a particular vegetation goal (diversity, cover, time). Should any of these parameters change, the recommendation would also probably change.

Natural colonization may be recommended for the entire site. In all cases where natural regeneration (all or in part) is to be applied, a realistic seed/propagule source must be present and have a realistic chance of establishing the vegetation on site. If so, the natural processes of dispersal, seedbank recruitment, and/or existing vegetation should provide sufficient propagules to successfully meet the revegetation goals. However, there are no guarantees. For example,

- An unexpected dispersal of noxious/invasive species occurs before the planned vegetation becomes established.
- An expected event does not occur on schedule.
- A flood or flow connection with an adjacent wetland that is intended to be the propagule donor source does not occur.
- The planned hydrology may not be realized during the establishment year and the intended vegetation does not establish.
- An unexpected rodent population infests the site.

There will always be the "what ifs," but problems that may occur need to be identified and incorporated into the monitoring and maintenance plan for the site.

Natural regeneration is not recommended for the site. When the keys lead to this recommendation, successful establishment by relying on natural recourses is doubtful based upon the project goals, objectives, or established success criteria. The planner has the responsibility to evaluate the recommendation on the basis of the intended project goals and expectations. Many methods of establishing vegetation by active means have been developed, and the planner should consider these alternatives.

Natural regeneration should occur within a specified linear distance. This recommendation limits the reliance on natural colonization to within a set linear distance from a propagule source that is expected to populate the site. These limits are based upon average dispersal abilities for woody and herbaceous vegetation and are provided as a general recommendation. These measures may be modified if the targeted vegetation has dispersal ranges outside the average.

The characteristics of each species that is intended to revegetate the site need to be evaluated as to its propagule dispersal pattern and longevity (live expectancy). Red maple (*Acer rubrum*) can be used as the first example. It is a wind dispersed, soft mast, woody plant. In the Southeast it sets seed and disperses in spring (April – May). The seed cannot withstand drying and must germinate within a few weeks of dispersal to survive. When relying on red maple to colonize a site, the site must be downwind of the seed source and be ready to receive the seed in spring when the seed load is dispersed.

Oaks (*Quercus* sp.) can serve as a different example. Most oak acorns are dispersed in the fall and have a longevity of 1 to 3 years depending on the species. Most oaks have no special adaptation for dispersal and cannot be expected to colonize areas beyond a few feet from the parent tree unless carried and planted by forgetful squirrels. However, overcup oak (*Quercus lyrata*) acorns float and are dispersed greater distances that most other oak species. To expect colonization by overcup, the site must be ready to receive the acorns in fall through winter and flooding from the parent source must occur.

An additional example is cottonwood (*Populus deltoides*). This species is wind dispersed in March to April and can travel distances much greater than the recommended distance for this category of no more than 100 meters. (Note: cottonwood seed remains viable for only 24 hours after the seed is shed). The "cottony" hairs on the seed that aid in wind dispersal also help the seed float. If cottonwood is not a target species and the site receives floodwater containing cottonwood seed, it will establish.

Wooded Wetland Decision Key

Natural Regeneration versus Active Revegetation

1. Hydrology and soil condition marginally altered onsite or significantly altered for less than 5 years .... go to 2
1. Hydrology and soil condition significantly altered onsite for more than 5 years .... go to B

    2. Propagules of desired species already exist onsite in adequate densities .... go to A
    2. Propagules do not exist onsite or do not occur in adequate densities .... go to 3

3. Desirable species occur onsite .... go to 4
3. Desirable species do not occur onsite .... go to 5

    4. Cover of plants is adequate to meet project objectives .... go to A
    4. Cover of plants is inadequate to meet project objectives .... go to 5

5. Restoration site is adjacent to a surrounding seed wall .... go to 6
5. Restoration site is not adjacent to a surrounding seed wall .... go to B

    6. Seed wall contains desirable species with hydrology tolerances similar to planned site conditions .... go to C
    6. Seed wall does not contain desirable species, or the species do not have hydrology tolerances similar to planned site conditions .... go to B

A. Natural regeneration may be recommended for the entire site.

B. Natural regeneration is not recommended for the site. Consider other methods to revegetate the site.

C. Natural regeneration should be no greater than 100 meters (about 300 ft) from the surrounding seed wall. Beyond this limit, consider other methods to revegetate the site.

Herbaceous Wetlands Decision Key

Natural Regeneration versus Active Revegetation

1. Vegetation already exists on site .... go to 2
1. Vegetation does not exist on site .... go to 4

    2. Desirable species occur on site .... go to 3
    2. Desirable species do not occur on site .... go to 4

3. Species diversity and cover is adequate to meet project objectives .... go to A
3. Species diversity and cover is not adequate to meet project objectives .... go to 4

    4. Site downstream, adjacent to, or near existing wetland .... go to 5
    4. Site not downstream, adjacent to, or near existing wetland .... go to 6

5. Adjacent wetland contains desirable species with hydrology tolerances similar to planned site conditions .... go to C
5. Adjacent wetland does not contain desirable species, or contains species with hydrology tolerances different from planned site conditions .... go to 6

    6. Wetland effectively drained less than 20 years .... go to 7
    6. Wetland effectively drained more than 20 years .... go to B

7. Seed bank contains desirable species with hydrology tolerances similar to planned site conditions .... go to 8
7. Seed bank does not contain desirable species or contains species with hydrology tolerances different from planned conditions .... go to B

    8. Density of seeds is adequate to meet project objectives .... go to A
    8. Density of seeds is inadequate to meet project objectives .... go to B

A. Natural regeneration may be recommended for the entire site.

B. Natural regeneration not recommended for site. Consider other methods to revegetate the site.

C. Limit natural regeneration to areas within 0.5 mile of emergent wetlands. Beyond this limit, consider other methods to revegetate the site.

To Contents

I.D.2 Active revegetation—utilizing donor sources

(Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, and John T. DeFazio, NRCS, New Albany, Mississippi, December 2001)

Purpose

This paper introduces several techniques that can be used for actively revegetating all or portions of a wetland restoration or enhancement site with materials derived from donor sites. These techniques rely on the use of donor wetlands as sources of plant materials and soil rich in seed and fleshy propagules. The processes of collecting and extracting donor materials and the introduction of these materials onto recipient wetlands to serve as the basis of revegetation are described. Topics include:

- Utilizing topsoils with propagules (mulching and inoculating),
- Using wetland sod mats and plugging donor soil,
- Making and using wetland hay.

Technical specifications on these techniques are included.

Contents

The restoration of vegetation is of critical concern when restoring or enhancing wetlands. Vegetation is the basis of food web support. It determines animal species diversity and abundance and is a critical factor in developing community structure. Depending on the site's conditions, in situ plant propagule sources, planned functions, and desired species, a wetland restoration or enhancement site may require some level of active revegetation to become properly vegetated for its intended purposes. Relying exclusively on recruitment from the propagule bank in the soil, or on seed immigrating onto the site, may result in limited species diversity and a site favoring species that are easily dispersed; i.e., noxious, invasive, and aggressive plant species (Burke 1997). A variety of successful methods has been developed that take advantage of local propagule sources found on adjacent wetlands and in their soils. Each method has applicability in different situations. Benefits can be maximized and success enhanced by combining some of these methods with others (Galatowitsch and van der Valk, 1994).

Definitions

Active revegetation—Establishing vegetation by physically placing seed, seedlings, cuttings, or other propagules onto a site. This includes a wide variety of activities ranging from completely covering a site with a suite of selected species to simply adding as few as one species to enhance a specific wetland function. Active revegetation often falls somewhere between these two extremes. For example, a common revegetation strategy is to establish a few dominant species, or species unlikely to disperse onto a site, while relying on natural regeneration processes to augment the remaining species diversity. Another example is applying plant materials to only the part of the site where the likelihood of colonization from natural sources is the most remote. In all cases, active revegetation means that there is some degree of adding plant materials to establish vegetation on the site.

Propagules—Seeds, seedlings, or fleshy plant parts (such as bulbs, rhizomes, cuttings, and pips) that are capable of establishing and growing into an adult. Depending upon the species, seed can stay viable in the soil propagule bank for an extended time, while fleshy propagules have a relatively short viability.

Propagule bank—Considered to be the viable seed, seedlings, and fleshy propagules contained in a soil. Various portions of the propagule bank (as defined here) have been considered separately by authors as seed bank, bud bank, seedling bank, and others depending on the type of material considered (Leck et al. 1989). However, for the purposes of this paper, propagule bank is inclusive of all types of plant propagules occurring in or on the soil.

Propagule bank characteristics

Characteristics of the propagule bank are important to consider when relying on propagules contained in a soil as a means of active revegetation on a restoration or enhancement site.

- Most seeds are found in the upper 2 inches of soil (Leck et al. 1989).

- Roots and fleshy propagules may extend down to 8 to10 inches (Galatowitsch and van der Valk 1994).
- As a rule, the seeds from weedy species and annuals have a longer longevity (life espectancy) than seeds of perennials.
- Soil contains seed and propagules of many different species, not only those actively growing on the site. They can be upland, wetland, native, alien, and invasive, regardless of the current hydrology characteristics of the site.
- Recent patterns of site use greatly influence both the quantity (numbers of a species) and composition (numbers of different species) of propagules in the bank (Brown 1998). For example, tillage depletes the seed of many species from a site while enhancing the quantity of others. Drained wetlands dominated by upland vegetation develop a seed bank proportionally high in upland (as compared to wetland) plant species.
- Dispersal of seeds from local sources predominate the seed concentration in the propagule bank, but dispersal from distant sources occur and may significantly affect the bank (Leck et al. 1989).
- Often the planned hydrology and the resultant hydrology of a restoration/enhancement site are not synonymous. The high diversity of species in donor soils can help in the establishment of vegetation on a newly restored site because species tolerant to the actual hydrology will respond (Burke 1997).
- Donor wetland soils contain organic material, fungi, and micro-organisms that are not found on many newly restored/enhanced sites. If the plant propagule bank is composed of species not suited to the site's hydrology, the addition of donor wetland soil is beneficial nonetheless (Burke 1997).

Collecting and storing donor topsoil

If the donor topsoil cannot be placed immediately into the restoration or enhancement site, the topsoil will need to be stockpiled. Stockpiling of wetland topsoil and its associated materials has had varied success. Because wetland topsoil contains the viable plant parts and seeds, these propagules may deteriorate from heat, freezing, aeration, desiccation, decomposition, or salt buildup during storage. Do not stockpile soil during summer, it will compost and kill both seed and fleshy propagules! However, be aware that composting can occur at any time of year. To maximize the chances for successful restoration and minimize composting impacts, stockpile soils in upland areas for less than 4 weeks. Donor soils consisting primarily of muck should be stored for no more than 2 weeks. Piles should be less than 3 by 3 feet (height/width) to avoid heat build-up. Periodical wetting of piles helps to cool the soil and prevents desiccation and loss of the fleshy propagules. Covering stockpiled soils with plastic sheeting may reduce drying and contamination with windborne weed seeds, but it can stimulate the heat buildup of composting resulting in seed/propagule deterioration.

Mulching and inoculating with donor topsoil

These techniques use the soil and plant propagule bank in the soil as the source of materials for revegetating a site. The source of soil and propagule bank can be derived from the restoration site itself (provided it has adequate topsoil and propagule bank), but is generally considered to be derived from other wetland sites. The level of intensity of adding the donor soil (mulching is most intense, followed by inoculating) is dependent on the restoration strategy, intended function or purpose, site characteristics, and budget. It is best to remove donor topsoil and re-spread while the plant materials are dormant (i.e., winter); otherwise, considerable losses occur.

Mulching is spreading the site with donor wetland soil up to 6 inches deep across the entire site or substantial portions of the site. Work the soil as little as possible to prevent mechanical damage to the seed and fleshy material. Typically, the quantities of soil needed in mulching are derived from a wetland site being impacted. Otherwise, the removal of the needed quantities of topsoil has too great an impact on the donor site.

Inoculating involves placing a thin layer of donor soil over the site or some portions of the site to augment the species diversity. Removal of small quantities of soil from the donor site causes minimal impact (but may be regulated locally). This technique is not intended to be the sole source of propagules for revegetation. Topsoil from upland sites may be used as a source of organic material and growing medium for vegetation. It should not be considered a propagule source for the wetland since the proportion of wetland plant materials in the propagule bank will be minimal.

Exhibit I.D.2–1 gives the technical specifications for mulching and inoculating with donor topsoil.

Sodmats and plugs of donor topsoil

These techniques both use pieces of soil with intact vegetation as a source of propagules introduced onto a restoration or enhancement site. The use of *sodmats* as a technique involves the removal of large sections of intact vegetation and soil from the donor site with almost immediate placement of the sodmat onto the receiving site. This technique can be done during most times of the year. It also leaves the plant roots and fleshy propagules intact with little to no disturbance. It does, however, destroy the donating site and is only recommended when the donating wetland is being impacted.

Using *plugs* of topsoil results in much less impact to the donating site. This technique involves the removal of small pieces of wetland soil, with its associated vegetation and propagule bank, and placement of the plug on the receiving site at a similar hydrology. Plugging will only diversify the restoration/enhancement site and is not intended to be the sole source of propagules for revegetation. The remainder of the vegetation will be derived from natural sources or by other methods if active revegetation.

Exhibit I.D.2–2 gives the technical specifications for sodmats and plugs.

Wetland hay

This technique involves the cutting of mature seed heads from wetlands, drying, bailing, and spreading the material on restored or enhanced sites. This technique allows for the targeting of specific species or a suite of species depending on the time of year the hay is collected. The seed from several wetlands and/or seed collected several times over a growing season from one wetland can be combined to increase species diversity. The hay can be harvested and stored for later use without losses as encountered by other methods using topsoils or sods. The removal of seeds from a wetland site for dispersal onto a restoration/enhancement site somewhat mimics natural dispersal methods and is less impacting on donor sites compared to other methods that involve removing the vegetation and soils. The removal of seed from a donor site affects the propagule bank, but large-scale impact can be avoided if the same sites are not used continually or the areas within a larger donor site are rotated in successive years.

Exhibit I.D.2–3 gives the technical specifications for wetland hay.

References

Brown, Stephen C. 1998. Remnant seed banks and vegetation a predictors of restored marsh vegetation. Canadian Journal of Botany, Vol. 76:620–629.

Burke, David J. 1997. Donor wetland soil promotes revegetation in wetland trials. Restoration and Management Notes 15:2, pp. 168–172.

Galatowitsch, Susan M., and Arnold G. van der Valk. 1994. Restoring prairie wetlands: an ecological approach. Iowa State University Press, Ames, Iowa, pp. 130–140.

Leck, Mary Allessio, V. Thomas Parker, and Robert L. Simson. 1989. Ecology of soil seed banks, Academic Press, New York, New York, 462 pp.

Exhibit I.D.2–1

Technical Specifications

Mulching and Inoculating With Donor Topsoil

Techniques

Mulching involves the removal of topsoil from a donor site and spreading it over the surface of a restored or enhanced wetland. Inoculating involves the removal of small amounts of topsoil from donor sites and spreading the donated topsoil thinly onto one or more small areas within the restored/enhanced wetland.

Method of establishment _____ **Mulching** _____ **Inoculating**

Mulching

Use a front-end loader to scrape the top 8 to 10 inches of soil from the donor wetland. Transport the donor soil to the project site by dump truck. Using a small bulldozer or scraper, spread the soil carefully over the substrate with minimal handling, overturning, or trampling. Spread the donor soil no more than 6 inches thick to prevent the seed and fleshy propagules from being buried too deeply. To ensure proper species placement, the donated topsoil should be placed at the same hydrology zone from where it was removed.

Inoculating

Remove a few cubic feet of topsoil from the donor wetland. To minimize impact to the donor site, remove no more than the top 2 inches of topsoil. To increase plant diversity on the receiving site, remove inoculating topsoil from several different donor wetlands in the area. Remove and stockpile topsoil from each vegetative zone separately. Re-spread the soil thinly (1 to 2 inches) on the receiving site at the same hydrology zone from where it was donated. Since changing environmental conditions favor some species over others in a given year, inoculations can be done over several years to maximize diversity. Caution—weeds and invasive vegetation may be a problem because the restoration/enhancement site will not be rapidly revegetated.

Timing

Mulching is best accomplished during late fall to early spring while the plants, seed, and fleshy propagules are dormant. If seed and other propagules are immature (fall) or have initiated germination (spring), success is greatly diminished.

Stockpiling

If donor soils cannot be spread immediately, stockpiled material is subject to composting. Stockpile in low volume piles (3 ft x 3 ft height/width, or smaller) to prevent heat build-up. Stockpile donor soils for no more than 4 weeks (2 weeks for muck soils). Periodical wetting of soil cools the pile and retards heat build-up. Do not stockpile soils in the summer or periods of high temperature.

Exhibit I.D.2–2

Technical Specifications

Sodmats and Plugs

Techniques
Large pieces or small plugs of wetland substrate from a donor wetland are placed into the wetland being restored or enhanced.

Method of establishment _____ **Sodmats** _____ **Plugs**

Collecting sodmats
A sodmat is a large, up to 8 foot square and 4 inches deep, piece of intact wetland soil and vegetation removed from a donor wetland site. It is cut from the donor wetland with shovels and a front-end loader modified with a sharp-edged steel plate that undercuts the sod for removal. The sodmat is loaded onto a flatbed truck for transport to the recipient wetland. Best results are achieved if the soils are moist, but well drained at the time of cutting. This reduces weight, helps the mat stay intact, and reduces "sticking" of the mat as it is being transferred on and off the transfer plate.

Placing sodmats
The sod pieces are placed in matching hydrological conditions from where they came and fit back together tightly in the same manner as sodding for a yard. Do not leave gaps between the sod mats. Invading weedy species will colonize the gaps. Since relatively large areas of the donor wetlands are impacted, this method should be used only as a salvage technique.

Collecting plugs
Plugs may be obtained using a coring device, such as a 4- to 6-inch diameter PVC pipe fitted with a handle, or other devise that maintains the integrity of the soil and living vegetation in the plug as much as possible (i.e., no soil augers). Remove enough soil in the plug to include plant roots of actively growing vegetation (about 4 to 6 inches). Individual plants and the associated soil can also be collected with a shovel and bucket. The weight of the plugs can quickly become a limiting factor. In addition, plugs may not remain intact during digging and transfer if there is not enough clay or organic matter to hold the plug together.

Placing plugs
Plugs can be planted with the same coring device used in their removal. The plug is placed into a newly formed hole and tamped well.

Timing
Sodmats and plugs from natural wetlands may be transplanted successfully at any time provided sufficient moisture is available in the recipient wetland to allow for continued growth and root development.

Exhibit I.D.2–3

Technical Specifications

Wetland Hay

Technique

This technique involves cutting and collecting mature vegetative material from a natural wetland and spreading the material on a restoration site. This technique has broad application and has been used successfully on wetlands and upland prairie restoration sites. It is best suited to emergent wetlands.

Collecting

The mature vegetation is clipped from a wetland by tractor with a side-mounted sicklebar, by hand, or by using another method to "lay the vegetation down" as opposed to chopping. This prevents shattering of the seed heads and makes collecting easier. Bales of barley or wheat straw are opened, spread-out linearly, and the wetland vegetation is spread onto the straw. Once dried, the straw and wetland vegetation is rebaled and stored for future use. Record the hydrology zone from which the wetland vegetation was derived and attach to the bale.

Method of spreading wetland hay onto receiving sites

After collection, spread the straw bales laced with wetland seed and vegetation onto the appropriate hydrologic zone of the restoration site. The straw and many wetland seeds will float and raft to the margins of the wetland. To prevent rafting, crimp the straw into to soil. To enhance species diversity, apply wetland hay from several marshes over a number of years.

Timing collection of donor sources

No exact season for clipping the donor wetland vegetation can be recommended. Because different species grow and mature over the course of a growing season, it is best to determine the vegetation type and species desired. Consult a botanical manual for the area if available (e.g., Michigan Flora); flowering and fruiting dates are generally listed in the species description. Then select the time of maturation of those species. Generally, late spring to mid summer clippings result in materials rich in sedges and rushes. Mid to late summer collections result in a high diversity of species. Late summer to fall clippings contain numerous composites and grasses. A secondary consideration for the time of clipping donor sites is the current site hydrology and the cutting equipment. It may not be possible to mechanically clip vegetation on sites that are excessively wet. The equipment will be detrimental to the donor wetland, and the clipped vegetation may fall into water and be lost. Removal of vegetation from the same sites over several years will adversely affect the propagule bank in the soil of the donor site. To prevent impact, rotate collection throughout the wetland or use multiple wetlands as donors.

I.D.3 Obtaining, storing, and propagating native wetland propagules

(Jennifer Kujawski, NRCS National Plant Materials Center, Beltsville, Maryland, December 2001)

Purpose

This paper provides information on the types of herbaceous and woody wetland materials that can be used for restoration and revegetation, how to obtain materials (whether by collection, propagation, or purchase), and how to store materials prior to use. Basic terms are defined in the text, and a list of reference materials for those interested in further reading is provided at the end.

Contents

Plant materials for wetland revegetation can be obtained in many ways. Each method has utility in some instances, but may not be the best choice for other projects. The process of choosing what plants will be used, in what form, and how they will be obtained should be thought out as far ahead of time as possible. The following criteria can help you make these decisions.

- Have a clear idea of the project goals and objectives. They can be as basic as restoring woody or herbaceous vegetation, or both, or as complex as determining the wetland functions aimed for—wildlife food and habitat, water quality improvement, or soil stabilization.

- Know the hydrology onsite. Certain plants tolerate certain water levels, and various types of plant materials can only be established under particular hydrologic regimes. It makes no sense to plant seeds of a moist soil sedge when there is standing water in an area—the seeds will not germinate, and even if they did, the plants would not tolerate those conditions.

- Know what other site factors are unique. Determine soil characteristics, if there is microtopography that can be exploited, if the site is shaded or full sun, and if there are animals like geese and deer that are a problem?

Once the list of potential species for the site is made, choose the appropriate plant form to use. Often, this decision is based on project budget and material cost. Seeds are usually less expensive to use than bareroot and container plants, but bareroot and container materials generally yield better (and more immediate) plantings.

Part of this choice of appropriate plant form depends on what is available. Along with this comes the issue of ecotypes. An ecotype is a population of plants that has become genetically differentiated in response to the conditions of a particular habitat and has a distinctive limit of tolerance to environmental factors (USDA NRCS 2000). For example, wetland plants growing around a pond in Maine will most likely have later flowering times and be more cold hardy than plants of the same species growing around a pond in Florida. When restoring wetland vegetation, consider using local ecotypes as much as possible. Some states may even require the use of local materials. Using plants that are already adapted to local conditions can contribute greatly to the success of a revegetation project.

Herbaceous plants

Herbaceous (non-woody) plants like grasses, sedges, rushes, and wildflowers are available in many forms, some of which can be readily assembled for a project. Several options are described here, but more detail is given on those options that can be reasonably accomplished by project participants.

Seed—Using seed to revegetate a wetland is often a low-cost technique, especially if the plan includes collecting the seed. Purchasing seed is more expensive than collecting, but presuming the collection was made by a professional, it ensures good quality seed and allows use of some unfamiliar species. Seeding a wetland can be tricky business since water levels must be carefully controlled during germination and establishment. Seed must remain in close contact with the soil surface to receive the three elements necessary for germination: moisture (not inundation), heat, and light (Hoag and Landis 2001). The chance for failure using seeds is greater than that for using plants for revegetation, and little information is available about direct seeding many species. However, seeding can be used in conjunction with other planting methods to enhance restoration.

If the plan is to collect seed of herbaceous plants, make sure that those collecting the plants can identify them and know when seed ripens. For example, soft rush *(Juncus effusus)* and fringed sedge *(Carex lurida)* are found in the same plant communities, but ripen at different periods of the year; more than one collection trip is necessary to catch both species when ripe. Regional floras list flowering and fruiting dates for the wetland species in that area. Consult these publications to get an idea of general collection windows and follow up with field observations. Check several times during the ripening period to determine the best time to collect—most seeds turn dark and become hard as they ripen.

Collection equipment needed includes a pair of hand clippers, bags (preferably paper because plastic bags can cause tightly packed seeds to overheat), and sturdy boots for wading. ***Note:*** collectors have had to use plastic bags for the *Juncus* spp. seed because it is so small that it falls right through the seams of the paper bags.

Collect plants from areas with conditions similar to the revegetation site, and be sure to get any necessary landowner permission or official permits before collecting. To maximize the amount of genetic diversity, try to collect from as many plants as possible, but do not strip a plant of all its seed. Seed collection ethics vary from species to species because some produce abundant seed while others produce few viable seed. A good rule of thumb is to collect no more than 10 percent of each plant's seed. Do not collect seed from rare, threatened, or endangered species.

Keep harvested seed dry and cool in the paper bags to prevent it from becoming moldy. Collected seed may need to be cleaned before use. Often seed is inside a protective fruit and will not germinate until the fruit is removed. Seed cleaning can be a relatively low-tech affair with hands and screens used to separate seed from chaff (table I.D.3–1).

If seed is purchased from a wetland vendor, look for high purity in the seed lot; that is, there should not be a lot of chaff, seed of other species, or damaged seed. If possible, get germination information. This information is not always done for wetland species other than grasses. It might be possible to get the results of a tetrazolium test, which is a test to determine the amount of seed able to germinate. ***Note:*** seed from a dealer will have a seed tag on it. The tag lists the percent germination, purity, weeds, other crop seed, and inert matter. If this information is not available from the dealer, it may be time to find a dealer who can supply this information. For more information, see Section I.D.5, Reading seed packaging labels, calculating seed mixtures, estimating cost.

Most herbaceous wetland seed, whether collected or purchased, requires some pregermination treatment before it will germinate. This treatment may be either stratification, a period of exposure to cold, moist conditions, or scarification, a treatment to make the seedcoat more permeable to water and gases. If seed is planted in the fall right after cleaning, winter temperatures and bacterial activity may take care of these requirements. If seed is held for spring planting, it can be rubbed with sandpaper lightly if scarification is needed, and should be stored cold (35 °F) in moist sand for 3 to 4 months to satisfy stratification.

Dormant propagules. Dormant propagules are overwintering, underground plant parts, such as rhizomes, bulbs, corms, and tubers. These parts are easy to work with and can be dug from wetland areas or purchased from vendors and transplanted into project sites. Revegetating a wetland with these materials is recommended over seeding because these propagules have more energy reserves to draw on than seeds. However, be aware these energy reserves are sought-after food for many wetland wildlife species.

If propagules are to be collected, dig as late as possible in the fall (but before the ground freezes, if this is an issue) to be sure plants are dormant. Be careful not to damage propagules as you dig since damaged parts can rot during storage and can actually degrade healthy propagules stored with them. If it is fairly certain that no noxious weeds are in the soil, some soil can be left around the propagules. This soil helps inoculate the new wetland with beneficial fungi and bacteria, and may provide seeds from new species. Be aware, however, that there is a risk of transporting

Table I.D.3–1 Seed processing and preplanting treatments for selected herbaceous wetland species

Species name	Ripening characteristics	Cleaning procedure	Preplanting treatment*
Arrowhead *(Sagittaria latifolia)*	Seedhead tan and falls apart when touched	Crumble seedheads between fingers to break up seeds	Stratify in moist peat for 3 to 4 months
Broadleaf, narrowleaf cattail *(Typha latifolia, T. angustifolia)*	Seedhead fluffy, brown	Pull seeds from stalk in a bag	None needed, store cold and dry until ready to seed
Ironweed *(Vernonia* spp.)	Tan fluff on top of gray-black seeds	Rub seedheads between fingers or over screen to break up clumps	None needed, store cold and dry until ready to seed
Northern blue flag, southern blue flag iris *(Iris versicolor, I. virginica)*	Large greenish-brown seed pods split open with brown, hard seeds inside	Pick seeds from pods	Stratify in moist peat for 3 to 4 months
Shallow sedge *(Carex lurida)*	Tan, papery covering over hard, brown seeds	Crumble seedhead between fingers to break up seed	Stratify in moist peat for 3 to 4 months
Soft rush *(Juncus effusus)*	Dry capsule containing dustlike, reddish-brown seeds	Crush capsules or shake over bowl to release seed	Stratify in moist sand or peat for 3 to 4 months for more rapid germination
Softstem bulrush *(Scirpus validus)*	Hard, dark-brown seed with light-tan hairs	Rub seed between fingers or over screen to break up seed cluster	Stratify in moist peat for 3 to 4 months
Switchgrass (*Panicum virgatum)*	Tan, hard seeds	Rub over screen to separate seeds from chaff	None needed, store cold and dry until ready to seed
Woolgrass *(Scirpus cyperinus)*	Reddish-brown seed in tangle of whitish "wool"	Rub wooly seeds over a screen and capture seeds below	Stratify in moist peat for 3 to 4 months

* Preplanting treatment is not necessary if seeding is done in the fall, soon after collection.

Source: Cape May Plant Materials Center 1997.

weed seeds, such as purple loosestrife *(Lythrum salicaria)* or phragmites *(Phragmites australis)*, with soil.

Store collected propagules in a cool, moist (not wet) location until needed. These materials have a much shorter shelf life than do seeds, so collection should be as close to planting time as possible.

Dormant propagules are also available from wetland plant vendors. As always, order from local sources when possible and order high quality material. If materials are purchased, they should be inspected upon delivery. Propagules should be firm, not mushy, and if they appear to be decomposing or smell bad, do not accept them.

In temperate regions, wetland plant materials require a cold treatment to break dormancy. Planting propagules during fall, winter, or early spring will ensure that they receive the cold period necessary to develop normally. Depending on the hydrology regime, frost heaving can be a major issue and lead to the loss of material. Tack the propagules into the substrate if possible. If frost heaving is suspected an issue, do not use dormant propagules unless they can be planted in the spring after the danger of frost heaving is past.

Donor wetland plugs. Wetland areas that are slated for destruction often provide the opportunity to save plants for a wetland revegetation project. Digging plugs can be done at any time of the year, although in hot weather, immediate transport to the project site or to cool storage is critical. As with any type of plant material collection, be sure to obtain permission before digging.

The basic tools needed to dig plugs from wetlands are a shovel, containers, and coolers (Hoag 1995). Be sure to dig as much of the root mass as possible. Plugs, particularly of some species of grasses, sedges, and rushes, can be divided into smaller clumps after digging. Transporting plants with soil may be advantageous in some cases (see dormant propagules), but increases the weight and bulk of the material to be transported to the new wetland. Once plants are dug, keep them cool and their roots wet. To keep transpiration low and allow plants to establish roots once they are planted in the new location, cut back plant tops on grasses, sedges, and rushes, but be sure to leave enough top growth to stick out of the water. Plant tops cut off below the anticipated water level can die from lack of oxygen (Bentrup and Hoag 1998).

Container plants. Using container plants to restore vegetation on a site can be costly, but healthy plants with intact root balls have an advantage over other plant materials. These plants do not need to expend energy on regrowing fine roots (as is the case with bareroot materials) or germinating and growing roots and shoots (as is the case with seeds and vegetative propagules). Container materials can be planted at any time of the year as long as the ground is not frozen and there is adequate moisture. ***Note:*** as with dormant propagules, frost heaving can be an issue in cold climates.

In most cases, propagating and growing container plants is done commercially; however if a project gets delayed, bareroot plants, dormant propagules, donor plugs, or even seeds that have limited longevity may need to be potted and stored. Most wetland plants are not particularly sensitive and do not require special soil. Clean topsoil is fine for most species. Soil that has a large amount of weed seeds can carry problem plants into the wetland. If clean topsoil is not available on site, bagged topsoil can be used. A 1:1 mix of sand and peat (or 1:1:1 sand, peat, perlite for better drainage) is also useful, especially for germinating small seeds of herbaceous species. While many wetland plants can grow under normal watering regimes, watering can be reduced and the plants can be acclimated to the intended site by letting containers sit in tubs partly filled with water. Let the water drain down before refilling the tubs to give the roots some oxygen and allow the plants to grow and spread much faster.

Most container plants are purchased from a wetland vendor. As with the other materials described, using a local supplier can help to minimize any difficulties the plants may have adapting to local climate conditions. Using local sources can also reduce the possibility of plants being damaged during shipment. Inspect container plants for overall health and appearance. Plant leaves should not appear pale or have yellowing or brown tips, and stems should be firm and flexible, not spindly and brittle. Look for evidence of pests or diseases, such as holes, wilting, or actual bug sightings. Pull plants from containers to look for strong root systems with many white roots. If specify sizes specified, be sure that the plant roots fill the containers. Herbaceous materials can be sold in various sizes, but most commonly as plugs, quarts, or gallon-sized containers. They are grown from seed, cuttings, vegetative propagules, or division. Containers may be plastic or biodegradable material, such as peat, paper, or fiber.

Woody plants

Woody plants for wetland revegetation are available in many of the same forms as herbaceous species; however, working with woody plants can take a bit more planning. Woody plants grow more slowly than herbaceous plants and several growing seasons are required for materials to be ready for transplanting.

Seed. The advantages and disadvantages to working with woody plant seed are similar to those for herbaceous seed. Using woody plant seed is generally inexpensive, but can be tricky, particularly with species whose seed is preferred animal food (e.g., acorns).

Woody plant seed vendors can provide seed for the project, but these suppliers are rare. Depending on the area of the country, seed of local origin can be difficult to obtain unless it is collected onsite. Viability or germination information is needed for any seed purchased.

Collecting seeds of woody plants requires a fair amount of logistics: locating several sources is essential since some plants do not produce reliable amounts of seed every year and the choicest seed is desired by birds and other animals. Two good sources of information for working with woody plant seed are *Seeds of Woody Plants in the United States* (USDA 1974) and *Seeds of Woody Plants in North America* (Young and Young 1992). These publications describe (species by species) seed ripening characteristics; collection, cleaning, and storage techniques; and preplanting requirements for many native trees and shrubs.

Before collecting seed from local sources, be sure to obtain permission from the landowners and keep in mind seed collection ethics (see Herbaceous plants section). A woody seed collection toolkit should include hand pruners, storage bags or buckets, a ladder to reach the fruiting branches of trees and taller shrubs (or a long pole or rake to knock ripe seed from branches), and a tarp to spread under plants whose seed can be shaken free. (A rifle or shotgun has proven useful in shooting off the upper branches of cottonwood and willow with their attached seed). Ideally, seed should be collected directly from trees or shrubs, but in some cases ground collection is the only method of access to the seed. Be aware that seed that has already dropped to the ground may be contaminated by soil pathogens, subject to predation by insects and rodents, and exposed to soil moisture that causes decay. Collecting freshly fallen seed less than a week old can reduce the possibility of contamination. Before collecting a large number of seeds, a cut test is a good way to check seed soundness. Cut open several seeds and look for a plump, firm, light-colored interior.

Some types of woody plant seed may require cleaning prior to planting or storage (table I.D.3–2). In most cases this can be accomplished by rubbing off outer coverings on screens; after rubbing, fleshy coverings can be washed away under running water. Sort out bad seeds with a float test: pour cleaned seeds into a bucket of water, stir, and remove seeds that float on the surface. Check a few floaters with a cut test to be sure they can be discarded. Air-dry sound seeds (those that sink) before storing or planting to avoid fungus growth. ***Note:*** sound acorns of overcup oak float as will seeds of willow and cottonwood, so be aware that the float test may not be useful for every species.

As with herbaceous seed, woody plant seed is best sown soon after collection. This will satisfy any stratification or scarification requirements. This recommendation is critical for some species; willow seed remains viable for about 7 days while cottonwood is viable for only 24 hours. It may also be possible, depending on species, to artificially scarify seed with files or sandpaper and hold seed in cold, moist storage for spring planting. Some woody plant seeds, particularly acorns, have a short shelf-life, and storage for longer than a few months results in reduced viability (see table I.D.3–2).

Hardwood cuttings. Stem cuttings made from woody plants during the dormant season are known as hardwood cuttings. These types of plant materials are particularly useful for revegetation on wetland edges and banks, above the water line. Cuttings are available to a limited extent from nurseries, but they are inexpensive, fast, and easy to prepare from local plant sources. Disadvantages to using hardwood cuttings are that they can dry out quickly and may have a high mortality rate, depending on site conditions.

Table I.D.3–2 Seed processing and preplanting treatments for selected woody wetland species

Species name	Ripening characteristics	Cleaning procedure	Preplanting treatment*
Blackgum *(Nyssa sylvatica)*	Fleshy fruit turns purplish-black	Rub fruit over sieve under running water to remove flesh from seed	Stratify in moist peat for 3 to 4 months
Buttonbush *(Cephalanthus occidentalis)*	Ball-shaped, reddish-brown fruitheads containing many 3 to 4 seeded nutlets	When dry, round fruitheads break up easily into separate nutlets (no need to separate individual seeds from nutlets)	None needed, store cold and dry until ready to seed
Sweetpepperbush *(Clethra alnifolia)*	Brown, dry capsules containing many tiny reddish-brown seeds	Shake seeds from capsules into a catch bowl	None needed, store cold and dry until ready to seed
Green ash *(Fraxinus pennsylvanica)*	Yellow-green or tan, winged seed	remove twigs, dirt, damaged seeds by hand	Sow immediately in fall after collection as seed needs both warm and cold stratification to germinate
Red maple *(Acer rubrum)*	Winged seeds are reddish-green, begin to drop from tree	Remove twigs, dirt, damaged seeds by hand	None needed, sow immediately after collection as seeds do not store well
Redosier dogwood *(Cornus sericea)*	While it is possible to collect the whitish fruits of dogwood, clean the pulp, and plant seeds after a 4-month stratification period, redosier dogwood is easily established with cuttings.		
Spicebush *(Lindera benzoin)*	Fleshy fruit turns bright red	Rub fruit over sieve under running water to remove flesh from seed	Sow immediately in fall after collection; seed does not store well
Swamp white oak *(Quercus bicolor)*	Acorns turn from green to brown	Remove defective acorns by hand or perform a float test (see text for details)	Sow immediately in fall after collection; seed does not store well
Willow *(Salix spp.)*	Planting cuttings is the best way to establish willows in a wetland. Willow seeds are viable only for a short period after collection.		

* Preplanting treatment is not necessary if seeding is done in the fall, soon after collection.
Sources: Young and Young 1992, USDA 1974.

Few species can actually sprout from hardwood cuttings. The most common exceptions are willow, poplar, and dogwood, which root readily without special treatment. After obtaining permission to collect cuttings from local populations, cut 1- to 3-year-old stems that are at lest 18 inches long and 1 to 1.25 inches in diameter (USDA 1998). This size recommendation may need to be modified when considering the local site conditions. For example, in the Western United States, the cuttings must be long enough to reach the lowest water table of the year. In high velocity situations, they need to be deeper to prevent dislodging by flowing water. In the high precipitation areas of the East, cuttings can be shorter, and thus more cuttings can be made from the donor material.

Hardwood cuttings should be stored in cold, dark locations that have high humidity until spring planting. Do not store them in moist conditions because the moisture encourages sprouting of roots along the entire stem. To prime cuttings to form roots quickly after planting, soak cuttings in water for 7 to 10 days before planting. This process swells the tissue that will expand from the cuttings to form roots.

Donor wetland plugs. Woody plant seedlings can be dug from impacted wetlands for revegetation use. Gathering these materials generally involves more work than seed or cutting collection because the plants require careful field digging and transport to the planting site.

Donor plugs of woody plants are best collected during the dormant season to avoid damaging the root systems of the plants. Be sure to dig as much of the root system of a plant as possible. Plants can be lifted with or without soil: there are advantages and disadvantages to either practice. Keeping the soil around plant roots can help transport beneficial organisms and seedbank to the new site, but also means heavier plugs to move. Digging plants bareroot makes transportation easier and reduces the risk of transporting undesirable weeds with the plants, but also means that the plants may dry out quickly.

Try to dig materials as close to the time they will be planted as possible. If storage is necessary, keep plants cool and moist.

Bareroot plants. Bareroot trees and shrubs are commonly grown by native plant nurseries and are fairly low cost materials to use. They are easy to store, transport, and plant, but survival is not as good as with materials that have the entire root system intact.

When purchasing bareroot plants, look for good quality seedlings. NRCS State foresters can provide information on the minimum stem length and root collar thickness for different species. Plants should have a substantial root mass left—about equal to the top. Do not accept materials that appear to have too much top growth to the amount of root. Plants should be firm, and the growing layer underneath the bark should be green when a small area of the bark is scratched (Environmental Concern 1997).

Store bareroot plants in a cool, damp, dark location. Moist sawdust or soil can be packed loosely around the plants to prevent the roots from drying out. Bareroot plants can be stored successfully for several months before planting as long as their roots do not dry out or freeze, and they are kept dormant to prevent leafing out.

Container plants and balled and burlapped material. Contained plants are the most expensive and cumbersome restoration materials, but also the most successful in terms of survival. Balled and burlapped (or B&B) plants are also expensive, but they can have lower survival rates because of the loss of roots when they are dug from nursery beds (similar to bareroot materials). Nursery procedure often cut the roots for B&B plants well before the time they are actually lifted. This stimulates additional root growth within the balled portion and alleviates some stress to the plant. Container and B&B plants can be planted at any time of the year as long as hydrology conditions are favorable and the ground is not frozen.

Because container materials for a project are rarely grown onsite, it is useful to know what to expect when trees and shrubs are purchased from commercial growers. Order early—talk to vendors as soon as it is known what is needed for the project. Propagation of woody plants, especially seedlings, can take two growing seasons or longer. Specify the plant size, not just the container size to avoid getting tiny plants in big containers.

Before accepting delivery of container or B&B stock, look at the quality of the materials, particularly the roots. With container plants, remove several plants from the pots and check roots to be sure they fill the pots and are large enough to support the top growth, but are not pot-bound. Large, thick roots circling inside the pots or girdling other roots indicate plants that have outgrown their containers and have not been transplanted to larger pots in time (Hoag 1997). B&B plants should have solid root balls with enough of the root systems present to support the top growth of the plants.

Overall quality is important. Plants for revegetation sites need not be perfect landscape specimens, but they should be vigorous and healthy, with no leaf damage, wilting, or insect pests (Environmental Concern 1997). Healthy plant material is most able to tolerate less than ideal conditions and survive on a restoration site.

Summary

Many plant material options are available for restoring or revegetating a wetland. The form or combination of forms best suited to a project depends on objectives, hydrology and other site conditions, project budget, and material availability. Local ecotypes of native plants should be chosen whenever possible because they are well adapted to the environmental conditions of the area. Forms of herbaceous plants useful for revegetation include seeds, vegetative propagules, donor wetland plugs, and container plants. Woody plant forms include seeds, hardwood cuttings, donor wetland plugs, bareroot, balled and burlapped, and container plants. Some of these materials are best purchased from wetland vendors, but others can be successfully collected from local sites. Whatever the form, it is important to use healthy plant materials in any planting project.

References

Bentrup, G., and J.C. Hoag. 1998. The practical streambank bioengineering guide: a user's guide for natural streambank stabilization techniques in the arid and semi-arid west. USDA NRCS Interagency Riparian/Wetland Plant Development Project, Plant Materials Center, Aberdeen, ID.

Cape May Plant Materials Center. 1997. Observations of wetland emergents. Unpublished data. USDA NRCS Plant Materials Center, Cape May, NJ.

Environmental Concern. 1997. Wetland planting techniques course handbook. Environmental Concern, St. Michaels, MD.

Hoag, J.C. 1995. Collection tips for herbaceous wetland plants. *In* View from a wetland: news and technology for riparian and wetland management, No. 2 (1995). USDA NRCS Plant Materials Center, Aberdeen, ID.

Hoag, J.C. 1997. Planning a project. USDA NRCS Riparian/Wetland Project Information Series No. 2, USDA NRCS Plant Materials Center, Aberdeen, ID.

Hoag, J.C., and T. Landis. 2001. Plant materials for riparian restoration. Native Plants J., Vol. 2. Issue 1, Jan. 2001.

United States Department of Agriculture, Natural Resources Conservation Service. 1998. Wetland restoration and enhancement student manual: bottomland hardwoods. National Employee Development Center, Fort Worth, TX.

United States Department of Agriculture, Natural Resources Conservation Service. 2000. National plant materials manual, 3rd ed., National Plant Materials Center, Beltsville, MD, 266 pp.

United States Department of Agriculture. 1974. Seeds of woody plants in the United States. C.S. Schopmeyer, ed., Agriculture Handbook No. 450, USDA Forest Service, Washington, DC, 883 pp.

Young, J.A., and C.G. Young. 1992. Seeds of woody plants in North America. Dioscorides Press, Portland, OR. 407 pp.

I.D.4 Soil seedbank assay technique

(Susan M. Galatowitsch, University of Minnesota, Department of Horticultural Science, December 2001)

Purpose

This section provides guidance on methodology useful in determining the plant species composition in the soil seedbank.

Contents

The viable seeds and other propagules that are present in the soil seedbank affects the vegetation on a developing wetland restoration or enhancement site. The quantity of seed, species diversity, proportions of each species, and presence of noxious or invasive species all influence the resulting vegetation. Knowledge of the seedbank characteristics for a site (or donor site) helps to determine a revegetation strategy. If some level of natural regeneration is planned as the means of revegetating a site, an understanding of the potential colonizers can help with planning decisions and maintenance concerns. The following information is provided as a method that has been successfully used to assess the soil seedbank on wetland restoration sites.

Equipment

The following equipment is needed:

- Soil collecting tool: a long-handled bulb planter, shovel, or soil auger.
- Soil sieve of 0.25-inch hardware or machine cloth (at least 12 by 12 inches) attached to a wooden frame or to a heavy plastic bucket with a cutout bottom.
- Container with an opening slightly smaller than the soil sieve.
- Plastic trays or flats without drain holes (at least 8- by 4-inch surface, 2 inches deep)—10 or more depending on wetland size.
- Well-lit, protected growing area (a greenhouse).

Procedure

Step 1. Collect surface soil or sediment (to a depth of about 3 inches) from the wetland. A long-handled bulb planter works well as a collecting tool in both natural and drained wetlands. Collect samples from around the wetland taking care to sample from each of the plant community assemblages. At least 20 locations at various hydrology levels should be sampled. These samples can be mixed together because seeds generally are well distributed across the hydrology zones of a wetland provided all parts of the wetland are contiguous and are managed in the same way.

- Seedbank samples are best collected early in spring when seeds are not actively germinating in the field. If the soil/sediment samples will not be used immediately, place the samples in sealed bags and keep cold (40 °F).
- Wetlands that are maintained separately from other wetlands are subdivided into different management units, or that are managed differently from other units must have the soil/sediment samples collected and processed separately.
- Wetlands over 100 acres need additional samples as well as those with considerable microtopographic relief and varied plant communities.

Step 2. Place the soil samples into the sieve and work it through the 0.25-inch hardware or machine cloth to remove roots and plant debris. The soil may need to be moistened with water and mixed to thick slurry so it can be sieved.

Step 3. Fill the growing trays with about 1.5 inches of a sterile soil medium (that has a pH comparable to the wetland soil). Spread sieved wetland sediment on top at a depth no more than 0.25 inch. This reduces ambiguity about whether plant seeds are buried too deeply to have the opportunity to germinate and assures that all of the seeds were close enough to the surface to do so. Place in a well-lit area protected from rain (trays will wash out during high-intensity rainfalls).

Step 4. Water once or twice daily, as necessary, to maintain saturated soil conditions. Do not water with a high-pressure hose that will dislodge newly germinated seeds from the soil or the soil from the tray.

Step 5. Periodically inspect the flats or trays for emerging wetland plants. Remove plants when they reach an identifiable stage. Fast-growing plants, especially grasses that tiller, need to be removed before they can be identified so they do not out-compete other seedlings. Carefully remove these plants with their roots, transplant to a separate pot, and continue to grow them out until they are identifiable.

Step 6. Maintain soil seedbank samples for at least 4 months to ensure most seeds that can germinate will do so. The few guides available for identifying seedlings focus on agronomic weeds. Often, plants must be grown to maturity to identify them with certainty.

I.D.5 Reading seed packaging labels and calculating seed mixtures

(J. Chris Hoag, USDA NRCS, Aberdeen Plant Materials Center, Aberdeen, Idaho, December 2001)

Purpose

This paper provides information on reading and identifying information on seed tags. It also provides guidance on how to determine the bulk seeding rate based on information from the seed tag, assistance in calculating seed mixes based on pure live seed (PLS) recommendations, and instructions on how to determine the best price per pound of different mixes based on PLS.

Contents

Seed lots vary widely in quality. Each lot of seed offered for sale to consumers is required by law to be properly and truthfully labeled; i.e., it must have a seed tag on it (fig. I.D.5–1). This applies to single species or a mixture, certified or noncertified. The information on the seed tag can help the user determine the quality of the seed lot. The bag can also have a certification tag on it (fig. I.D.5–2). Certification tags are in addition to the actual seed tag and establish that the seed meets the standards set out for each certified class of seed.

Rarely should agencies or Federal landowners recommend purchasing anything but certified seed. The use of certified seed provides the genetic and mechanical purity and varietal identity needed to establish a uniform seeding and minimizes the risk of introducing weed seed.

Figure I.D.5–1 Example of a seed tag from the USDA NRCS Plant Materials Center, Aberdeen, Idaho

Species ______________________ Acc. no. __________

Commmon name ______________________ Year grown __________

Weight ______________________ Origin ______________________

Purity ______________________ % Germination ______________________ %

Other crop seed ______________________ % Hard seeds ______________________ %

Total germination ______________________

Inert Matter ______________________ % and hard seeds ______________________ %

Weed seeds ______________________ % Date of test ______________________

Noxious weed seed ______________________

This seed was produced, collected or purchased by the
U. S. Government for use in conservation plantings
Aberdeen Plant Materials Center-Soil Conservation service
United States Department of Agriculture, Aberdeen, Idaho

Information on a seed tag:
- Variety and kind (species and common name)
- Lot number
- Origin
- Net weight
- Percent pure seed
- Percent germination (and date of test)
- Percent inert matter
- Percent other crop seed
- Percent weed seeds
- Name of restricted noxious seed (number per pound of seed)
- Prohibited noxious seeds are not allowed.
- Name and address of company responsible for analysis (seller)

The Federal Seed Act and State seed laws dictate the information found on the seed tag. Certification agencies of all States comply with the minimum requirements and standards of Association of Official Seed Certification Agencies (AOSCA). Additional information can vary slightly from State to State. Variety and kind (species and common name), lot number, origin, and net weight are all obtained from the grower or seed conditioner (seed cleaner). The remaining information is obtained from the Seed Analysis Report (fig. I.D.5–3 and I.D.5–4). An official seed laboratory completes this report. These labs can be either governmental, commercial, or private. The Seed Analysis Report lists all the seeds found in the test sample lot. With the Seed Analysis Report in hand, the buyer, not the seed dealer, decides what is to be seeded. It is the buyer's right to receive the Seed Analysis Report. If certified seed that is properly labeled is purchased from an adjacent State and if the Seed Analysis Report is available, buyers can count on the seed being similar in quality to their own State's certified seed standards because there are only small differences in seed standards between States. The seed lab performs a number of tests on a representative sample from each lot that is submitted by State or certification officials, the grower, seed conditioner, or seed dealer. The tests are conducted under controlled conditions based on the Rules for Testing Seed adopted by the Association of Official Seed Analysts.

Figure I.D.5–2 Example certification tags

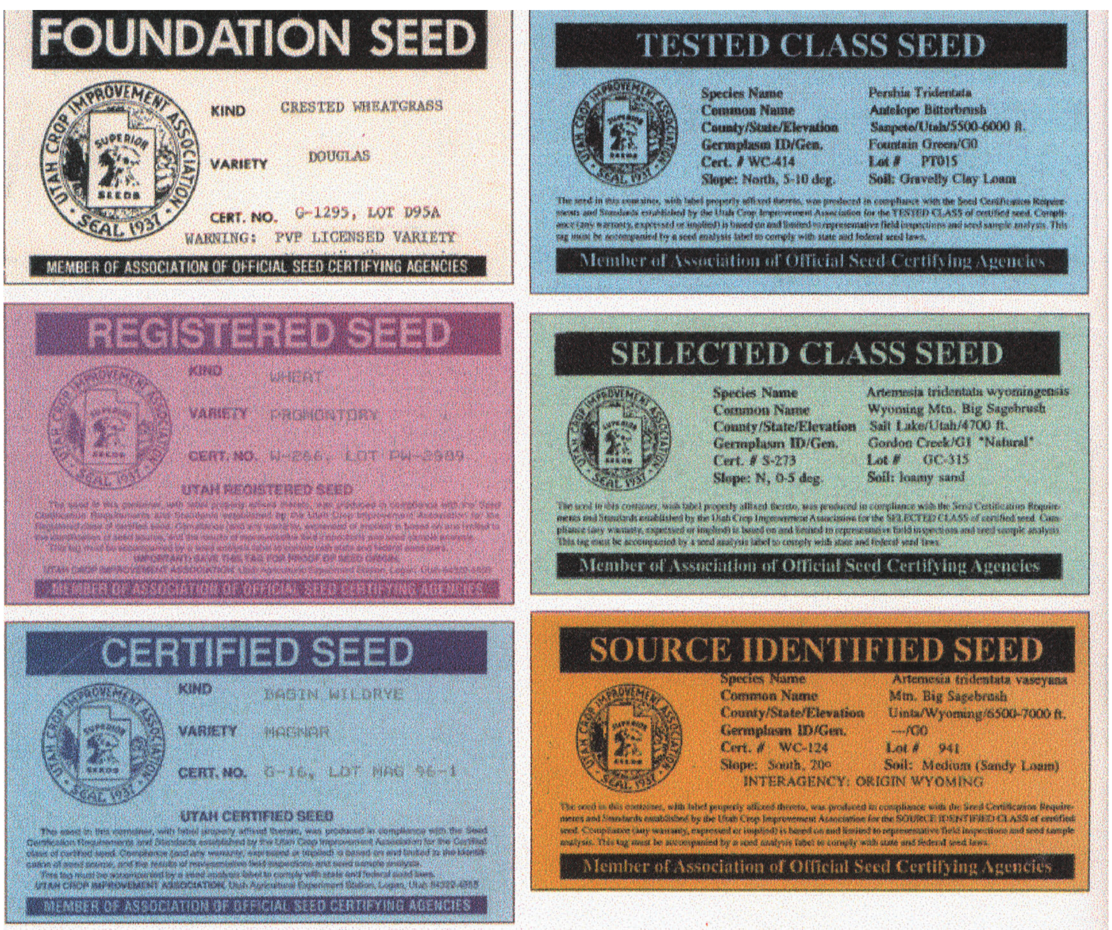


Figure I.D.5–3 Example of a seed analysis report issued by the Idaho Department of Agriculture, Idaho State Seed Laboratory, for bluebunch wheatgrass (note common weed seeds listed with the number of seeds per pound found)

STATE OF IDAHO — DEPARTMENT OF AGRICULTURE
IDAHO STATE SEED LABORATORY
2240 Kellogg Lane • Boise, Idaho 83712
Telephone: 334-2368

FINAL REPORT

RECEIVED APR 1996

Test Number	Date Received	Date Reported	Sampler	Sample Received From
96-08536	03/05/96	04/01/96		Plant Materials Center

Lot	Sender's Identification	To Be Tested As
PMC-95-2-B-10 BIN 10	Bluebunch wheatgrass, Goldar	Foundation

Grower Name and Number | Container | Pounds

TO

Plant Materials Center
Soil Conservation Service
Box 296
Aberdeen, ID 83210-0296

TEST REQUESTED

PURITY GERMINATION
IDAHO NOXIOUS

TEST RESULTS

Purity Based On 7.0000 Grams — Germination Based On 400 Seeds — Date of Germination 04/01/96

KIND OF SEED	% Pure Seed	% Other Crop	% Inert Matter	% Weed Seed	% Germ	% Hard Seed	% Dormant Seed	% Tetrazolium	% Hard Seed
Bluebunch wheatgrass (Pseudoroegneria spicata) Goldar	95.75	0.11	3.88	0.26	77	0			

INERT MATTER CONTAINS Ergot, Stems, Chaff

CROP SEEDS	No. Per Pound	% By Weight	WEED SEEDS	No. Per Pound	% By Weight
Slender Wheatgrass Elymus trachycaulus	130		Flixweed Descurainia sophia	130	
			Meadow salsify Tragopogon pratensis	65	
			Downy brome Bromus tectorum L.	194	
			Vervain Verbena sp.	518	
			*** IDAHO NOXIOUS *** NONE FOUND 74.56 GRAMS EXAMINED ***		

REMARKS:

Test results indicate this sample does not qualify as Foundation.
ICIA will determine final class on all certified samples.

Richard C. Lawson
Richard C. Lawson, Chief
Bureau of Seed Analysis & Control

Information contained in this report is based upon the sample as received from the sender. The State Department of Agriculture makes no warranty as to the variety of this seed. The Noxious exam conducted is Idaho State only unless otherwise specified.

Figure I.D.5–4 Example of a seed analysis report issued by the Idaho Department of Agriculture, Idaho State Seed Laboratory for Indian ricegrass (note common weed seeds and one noxious weed species detailed on the sheet)

STATE OF IDAHO — DEPARTMENT OF AGRICULTURE

IDAHO STATE SEED LABORATORY

2240 Kellogg Lane • Boise, Idaho 83712

Telephone: 334-2368

CECIL D. ANDRUS
Governor

RICHARD R. RUSH
Director

FINAL REPORT

Test Number	Date Received	Date Reported	Sampler	Sample Received As
90-08324	03/23/90	03/28/90		UNCERTIFIED

Lot	Sender's Identification	Class
SFP-BL-8-6 BIN 6	Indian Ricegrass, Nezpar, Blend	

Grower Name and Number	Container	Pounds

TO

Plant Materials Center
Soil Conservation Service
Box AA
Aberdeen, ID 83210

TEST REQUESTED

PURITY TETRAZOLIUM
IDAHO NOXIOUS

TEST RESULTS

KIND OF SEED	Purity Based On 7.0000 Grams				Germination Based On Seeds				Date of Germination
	% Pure Seed	% Other Crop	% Inert Matter	% Weed Seed	% Germ	% Hard Seed	% Dormant Seed	% Tetrazolium	% Hard Seed
Indian Ricegrass (Oryzopsis hymenoides)	93.89	0.00	5.74	0.37				97	0

INERT MATTER CONTAINS Broken Seed, Stones, Stems

CROP SEEDS	No. Per Pound	% By Weight	WEED SEEDS	No. Per Pound	% By Weight
			Mustard, Blue * Chorispora tenella	648	
			Mallow, Common Malva neglecta	259	
			Witchgrass Panicum capillare	65	
			Barnyardgrass Echinochloa crusgalli	194	
			Dock, Curly Rumex crispus	65	
			*** IDAHO NOXIOUS *** 70.55 GRAMS EXAMINED ***		
			Cress, Hoary Cardaria araba	6	

REMARKS: Purity - 13.50
TZ Germ - 20.00
Total 33.50

Richard C. Lawson
Richard C. Lawson, Chief
Bureau of Seed Analysis & Control

Certification Grade based on Laboratory Test. Final and official certification Grade to be determined by Idaho Crop Improvement Assoc. Inc.	Final Class	Rejected	Reason

Information contained in this report is based upon the sample as received from the sender. The State Department of Agriculture makes no warranty as to the variety of this seed. The Noxious exam conducted is Idaho State only unless otherwise specified.

A purity test separates pure seed, inert matter, other crop seed, and weed seed.

- **Purity** expresses the composition of the seed lot and its degree of contamination by unwanted components.
- **Inert matter** includes soil, plant parts, and certain types of damaged seeds.
- **Other crop seeds** are those species normally grown for crops that occur in amounts of 5 percent or less.
- **Weed seeds** gives the total amount of common and restricted weed seed found in the lot. *Note:* the number of noxious weed seeds per pound is listed separately.
- **Percent germination** gives the result of a germination test that determines the capability of a seed lot to produce normal seedlings under favorable, controlled conditions. Total germination is the percent germination added to the percent hard seed. Anything under 100 percent total germination represents the presence of dead seed and/or seed that does not produce a shoot or root.
- **Dormant seed**, which includes hard seed, is normally associated with legumes. It refers to the portion of the seed sample that does not germinate during the seed evaluation. Reasons for dormant seed are the seed coat is impervious to water and internal structures within the seed prohibit oxygen exchange. Hard seed has a seed coat that is impervious to water. This seed may germinate later and produce a viable plant, germinate and succumb to competition, or never germinate at all.

In addition, the lot sample is examined for the presence of restricted or prohibited weed seed. Each State has its own Prohibited and Restricted Noxious Weeds list. Restricted weeds in the originating State may be different from those in the receiving State. The lists are assembled from input provided by seed growers, crop improvement associations, seed dealers, and others. They do not necessarily include all the weeds all groups considered noxious.

By the Federal Seed Act and State law, seed bags cannot be sold when they contain any prohibited noxious weed seeds. In addition, seed bags can contain only a small percentage of restricted noxious weed seeds that are listed as the number of seeds per pound. This means that any one of these weed seeds on the restricted list can be included in a certified seed bag up to the maximum number allowed by law. Even with only a few seeds per pound, there are more than enough weeds to infest thousands of acres.

Common weed seeds are those weeds that are not on the Prohibited or Restricted Noxious Weed lists. Common weeds can be included in higher amounts in some States. The major problem with common weeds is that they are not listed on the tag, so buyers do not know what they are. The only way to know what they are is from the seed analysis report.

Prohibited and restricted weed lists vary from State to State; however, each state requires similar information. Identify the State's requirements to know what the buyer is legally entitled to or legally obligated to do. By law, seed that comes from a State that has less restrictive weed lists must meet the more restrictive requirements of the receiving State.

The use of certified seed helps protect the buyer. Certified seed is the best quality because it has to meet specific standards of high genetic purity, germplasm identity, high germinating ability, and minimum amounts of other crop seed, weed seed, and inert matter. A clear understanding of the certified seed standards for each State is critical to know what can be found in many seeds.

AOSCA has recently published pre-varietal germplasm certification standards for the certification of germplasm accessions that have not been released as a variety. These standards offer a reliable way for the seed industry to offer seed of varieties, races, or ecotypes to the buyer that still has genetic identity, but they do not go through as extensive testing as a variety does. This means that seed of plants that are released under the alternative release procedures can get to the field much faster. These new release procedures are most often used for native species.

Calculating seed mixtures

All NRCS recommendations are expressed in pounds of pure live seed (PLS). PLS is defined as the percentage of pure seed that will germinate expressed as a percentage of a given weight of seed. It provides a common basis for comparing seed lots that differ in purity and germination. It is also used to adjust seeding rates to achieve maximum production after seeding. The basic formula to calculate PLS is

$$\text{PLS} = \frac{\text{percent purity} \times \text{percent germination}}{100}$$

The information necessary to complete this calculation is found on the seed tag or the seed analysis report. The following example shows how PLS is calculated for one lot of Goldar bluebunch wheatgrass on which the seed tag indicates 99.01 percent purity and 87 percent germination.

$$\text{PLS} = \frac{(99.01)(87)}{100}$$
$$\text{PLS} = 86.13\%$$

Once PLS is determined for the lot of seed, it can be used to compare the seed costs of two different priced seed. An example cost analysis for using alfalfa is shown in example I.D.5–1.

Example I.D.5–1 Cost comparison for two different priced alfalfa seed

Given: Dealer X and Dealer Y have the same variety of alfalfa for sale.
Dealer Y's alfalfa seed is selling for $.90 per pound. The seed analysis report lists the purity as 99.5% and the germination as 90%. The percent PLS is 0.8955. Dealer X's alfalfa seed is selling for $.70 per pound. The seed analysis report lists the purity as 93.0% and the germination as 60%. The percent PLS is 0.5588.

Determine: The better deal.

Solution: Use the following formula for calculation of both dealers' price:

$$\text{Price per pound (PLS)} = \frac{\text{price per pound}}{\text{percent PLS}}$$

Dealer Y:

$$\text{Price per pound (PLS)} = \frac{\$90}{0.8955}$$
$$\text{Price per pound (PLS)} = \$1.01$$

Dealer X:

$$\text{Price per pound (PLS)} = \frac{\$70}{0.5588}$$
$$\text{Price per pound (PLS)} = \$1.25$$

From these calculations, it is easy to see that the posted price is not always the cheapest.

Adjusting seeding rates

To seed the recommended PLS seeding rate, the bulk rate of seeding needs to be determined. The drill will be set at this rate since the other material in the seed lot cannot be removed. This bulk seeding rate is always higher than the PLS seeding rate. The formula to calculate the bulk seeding rate is as follows:

$$\text{Pounds bulk seeding rate per acre} = \frac{\text{pounds PLS recommended rate per acre}}{\text{percent PLS}}$$

An example of how this formula is used follows:

The NRCS recommended seeding rate for Hycrest crested wheatgrass is 7 pounds PLS per acre. The PLS is calculated to be 80 percent. The bulk rate needed to seed the recommended PLS rate is determined by:

$$\text{Pounds bulk seeding rate per acre} = \frac{\text{7 lb PLS recommended rate per acre}}{\text{0.80 PLS}}$$

Bulk seeding rate per acre = 8.75 pounds

Based on these calculations, the drill box setting would be as close to 8.75 pounds per acre as the model of drill will allow.

Seeding rates for mixtures

Where a seed mix will be used, the percent of each species desired in the mixture needs to be determined. To do this, multiply the percent desired in the seed mix times the pounds of PLS recommended per acre to get the PLS mix per acre. Example I.D.5–2 shows the calculation of seeding rates for mixed seed.

Example I.D.5–2 Calculation of seeding rates for mixed seed

Given: Of the desired seed mix, 85% will be Goldar bluebunch wheatgrass. This lot of seed has a 90% PLS. The NRCS recommended seeding rate is 8 lb PLS/acre. The remaining 15% of the mix will be Delar small burnet. This lot of seed has an 85% PLS. The NRCS recommended seeding rate is 20 lb PLS/acre.

(Goldar 85%) (8 lb PLS/ac) = 6.8 lb PLS/ac mixed

(Delar 15%) (20 lb PLS/ac) = 3.0 lb PLS/ac mixed

Determine: Amount of bulk seed (mixed) per acre using the formula as explained above.

Solution:

$$\text{Goldar } \frac{\text{6.8 lb PLS /ac}}{90\%} = \text{7.6 lb bulk mixed/ac}$$

$$\text{Delar } \frac{\text{3.0 lb PLS /ac}}{85\%} = \text{3.5 lb bulk mixed/ac}$$

Multiply the pounds of bulk seed per acre for each species by the acres to be seeded to obtain the total bulk seed required for the entire seeding project acreage.

Recommendations

Information on the seed label and in the seed analysis report can be used to determine the quality of the seed that is being purchased. This in turn will ensure that genetic and mechanical purity, in addition to varietal identity, needed to ensure a successful, weed-free, uniform seeding can be accomplished. Purity and germination percentages found on the seed tag can help to determine pure live seed from which the bulk seeding rate can be determined. The seed tag and seed analysis report also lists the weeds in the seed lot including common, restricted, and prohibited weed seeds. Remember that weeds listed as common, restricted, and prohibited vary by State. Seed that is moved across State lines must meet the most restrictive State's requirements. Monitoring the weed species in the lot can help to control what weeds are seeded in a planting.

The cheapest seed is not always the most economical. Comparing the purity and germination percentage between seed lots or mixes clearly shows which lots or mixes will produce the most seedlings after planting. All seeding recommendations are given in Pure Live Seed rates. These rates must be converted into bulk seeding rates before the seed is placed in a drill to begin seeding. The drill must also be set up based on bulk seeding rates, not Pure Live Seed rates. Always place seed orders to the seed dealer as bulk seeding rate and check to make sure the dealer has mixed the seed mix correctly.

Literature cited

Association of Official Seed Certifying Agencies. 1999. Genetic and crop standards manual. Boise, ID.

Idaho Crop Improvement Association. 1998. Idaho rules of certification. Meridian, ID.

United States Department of Agriculture, Natural Resources Conservation Service. 1998. Idaho pure seed law and rules and regulations—pure seed law. Plant Materials Technical Note No. 21, Boise, ID.

Additional resources

Dickerson, J., and B. Wark. 1999. Vegetating with native grasses in Northeastern North America. USDA Natural Resourc. Conserv. Serv. and D Univ., Syracuse, NY.

Hoag, J.C., S.K. Wyman, G. Bentrup, D.G. Ogle, J. Carleton, F. Berg, and B. Leinard. 2000. Users guide to description, propagation, and establishment of sedges, rushes and grasses for riparian areas in the Intermountain West. USDA Natural Resourc. Conserv. Serv., Idaho and Montana Technical Note, Aberdeen, ID, and Bozeman, MT.

Ogle, D.G., and J.C. Hoag. 2000. Stormwater plant materials, a resource guide. City of Boise Public Works Dep., Boise, ID.

Stevens, R., K.R. Jorgensen, S.A. Young, and S.B. Monsen. 1996. Forb and shrub seed production guide for Utah. Utah State Univ. Ext. Serv., Logan, UT.

United States Department of Agriculture, Natural Resources Conservation Service. 1998. Improved grass, forb, legume, and woody seed species for the Intermountain West. Plant Materials Technical Note No. 24, Boise, ID.

United States Department of Agriculture, Natural Resources Conservation Service, 2000. National plant materials manual, 3rd ed. Beltsville, MD.

To Contents

I.D.7a Restoring herbaceous wetland vegetation by seedlings

(J. Chris Hoag, NRCS Plant Materials Center, Aberdeen, Idaho, December 2001)

Purpose

Information in this section provides planting techniques to allow successful restoration of herbaceous wetland vegetation. Wetland species are notoriously difficult to seed. Using plugs, either greenhouse grown or wild transplants (wildlings), is often the only successful method available. This paper gives techniques for collecting, propagating, and transplanting wetland plant seeds and wildlings for wetland revegetation projects

Contents

Sedges (*Carex* spp.), spikerushes (*Eleocharis* spp.), bulrushes (*Scirpus* spp.), and rushes (*Juncus* spp.) are used extensively in riparian and wetland revegetation because of their aggressive root systems (fig. I.D.7a–1). They also provide wildlife habitat for a variety of terrestrial and aquatic species and form buffer zones that remove pollutants from surface runoff. The aboveground biomass provides roughness that causes stream velocity to decrease and sedimentation to occur. The thick humus developing in those areas breaks down organic compounds and captures nutrients (Carlson 1992).

Figure I.D.7a–1 Aggressive root system and rhizomes

Wetland plant root systems are important means of stabilizing degraded sites. Manning et al. (1989) found that Nebraska sedge (*Carex nebrascensis* Dewey) produced 212 feet per cubic inch (382.3 cm/cm^3) of roots in the top 16 inches (41 cm) of the soil profile, and Baltic rush (*Juncus balticus* Willd) had 72 feet per cubic inch (134.6 cm/cm^3) of roots. An upland grass like Nevada bluegrass only has 19 feet per cubic inch (35.3 cm/cm^3) of roots. The root system is the basis for soil bioengineering. Soil bioengineering increases the strength and structure of the soil and thereby reduces streambank erosion. Most soil bioengineering applications emphasize the use of woody riparian plants. However, herbaceous wetland plants provide more fibrous root systems that in combination with the larger woody plant roots do a better job of tying the soil together (Bentrup and Hoag 1998).

Wetland plants are also used for constructed wetland systems (CWS). A CWS is a wetland that is constructed in an area that has no previous history of wetland hydrology for the purpose of improving water quality. Water purification is a natural function of wetlands. The wetland plants provide suitable sites on which colonizing microbial populations can establish. The microbial populations live on the plant roots and break down various nutrients found in the water. The aboveground biomass serves as a nursery site for periphyton that also break down various nutrients.

Direct seeding of wetland plants

Many wetland plants are difficult to seed in the wild. Wetland plant seeds generally need three things to germinate: heat, water, and light. The need for light means that wetland plant seeds need to be seeded on the surface and they cannot be covered with soil (Grelsson and Nilsson 1991, Leck 1989, Salisbury 1970). Drilling the seed with a drill covers the seed especially if packer wheels or drag chains are used.

Many species have a hard seed coat that takes a year or longer to break down enough for the embryo to germinate. Many species require special stratification treatments to prepare the seed for planting. These treatments include everything from acid wash to mechanical scarification, from prechilling to extremely high temperature soil conditions. Occasionally, dormant seeding (seeding during late fall or winter after the plants have gone dormant) can be successful, but it depends on the species.

Not having absolute control of the water going into the wetland or riparian area is the most common mistake that occurs when seeding wetland plants. Without good water control, when water enters the system the newly planted seeds float to the water surface and move to the water's edge where wave action deposits the seed in a narrow zone. The seed germinates here, and the stand is generally quite successful as long as the hydrologic conditions are maintained for the various species deposited there (Hoag et al. 1995). With good water control, the seeds generally stay in place and the stand covers the wetland bottom instead of just around the fringe.

Some species when seeded in a greenhouse setting need a cold-hot stratification environment for successful germination. This means that the seeds are placed in cold storage at 32 to 36 degrees Fahrenheit for 30 to 60 days and then they are planted in moist soil containers at about 100 degrees Fahrenheit. Heat is one of the essential requirements for germination and growth. (Hoag et al. 1995)

Based on these difficulties, using direct seeding of herbaceous plants as the primary means of revegetating a site requires more attention to planning and control of site hydrology during the establishment period to be successful. It also requires knowledge of the specific germination/stratification requirements (if any) of the targeted species. Successful establishment of herbaceous vegetation by direct seeding is possible and examples of these successes range from the establishment of tufted hairgrass (*Deschampsia caespitosa*) wetlands in Oregon to multiple species herbaceous depression wetlands in Delaware. Typically, however, direct seeding of herbaceous species is not used as the primary means of active revegetation, but it is a method to increase the overall species diversity in a wetland, especially around the perimeter, and to establish populations of specific target species.

Revegetating a site with herbaceous species plugs of greenhouse-grown material has shown a much higher establishment rate than with seeding or collections of wildlings (Hoag et al. 1995). The rest of this section describes the use of wetland plants as a means of actively revegetating herbaceous vegetation on restored and enhanced wetlands.

Collection and propagation of wetland plants

Woody shrubs, grasses, and wetland plants are often grown in small containers or plugs [volumes less than 22 cubic inches (361 cm^3)]. Plugs are used in bioengineering designs when the water is too deep or persistent to get woody plants established in other ways (fig. I.D.7a–2). Transplanting wild plants (wildlings) (fig. I.D.7a–3) is sometimes used, but small-volume

Figure I.D.7a–2 Herbaceous material plug

Figure I.D.7a–3 Collecting wildling from an existing wetland containers often have higher establishment rates and spread faster and further (Hoag 1994). The two basic procedures for obtaining wetland plant plugs are growing them or harvesting wildlings from a donor site.

Greenhouse propagation

As previously stated, water, heat, and light are required to grow wetland plants from seed. The need for water is fairly straightforward especially when the conditions in a natural wetland are considered. Light, however, is not as obvious. Covering wetland plant seeds with even a thin covering of soil significantly decreases germination of some species. Heat is also less obvious. Natural wetlands are generally hot and humid. Research has found that greenhouse temperatures of 100 degrees Fahrenheit or higher increase germination and growth.

Seeds of most of the wetland plants except rushes need to be stratified. Stratification is essentially "fooling" the seeds into germination mode by mimicking the environmental conditions that they would be subject to had they remained outside during the winter. The seeds are stratified in small, plastic containers that are filled with distilled water and have 0.3 ounce (8 g) of loose sphagnum moss added to the water in the bottom of the container. The seeds are put into a coffee filter, and the filter is nestled down into the moss. The containers are placed in a dark cooler for 30 days at 32 to 36 degrees Fahrenheit. At the end of 30 days, the seeds are removed from the stratification medium.

Special propagation tanks and Rootrainers™ with a 1:1:1 soil mix of sand, vermiculite, and peat are used when planting wetland plant seeds in the green house. Rootrainers™ have a large hole in the bottom that needs to be covered so the soil does not wash out when water is added to the tanks. A single sheet of paper towel crumpled up and shoved into the mouth of each cell will prevent this. The seeds are placed on the soil surface of the cells in each Rootrainers™ after the surface has been firmly packed. A 2- by 2-inch (5- by 5-cm) wooden tamp works well and can pack the soil to a sufficient density that a finger barely makes an impression in the soil surface. From 5 to 10 seeds are put on a finger and pushed onto the soil surface. The seeds need to be in good contact with the soil surface.

After the stratified seeds are planted on the soil surface, the tanks are filled with water to within about 1 inch of the soil surface. The seeds should be illuminated for 24 hours a day with 400-watt metal halide lamps for the first month. The lights can be turned off after 1 month. Covering the propagation tanks with clear plastic while the seeds are germinating helps keep the environment warm and humid. If damping off of the seedlings is a problem, try flooding the soil. Leave the soil completely submerged under 0.25 to 0.5 inch (6.4 to 12.7 mm) of water for about 2 weeks. After this period lower the water level. This procedure will subdue the fungus and may stimulate more stubborn seeds to germinate. Do not flood the soil if the seeds have not germinated or they will float and move out of the cells.

With this method, 22-cubic-inch (361 cm^3) plants can be grown from collection to full size in less than 100 days. Plugs can be held in the greenhouse if necessary for extended periods with minimal maintenance. Several crops can be raised throughout the year because of the short turnaround time.

If growing the plants is not an option and they must be purchased, the following concerns need to be considered.

- The grower needs to be willing and able to grow wetland plants that can be difficult to propagate.
- The grower must understand the special propagation requirements and be able to accomplish them.
- The grower must understand the project plant requirements in terms of height and size at the time that the contract is signed.

Determine the planting date before going to the grower so that he/she knows when the plants need to be ready. Check in with the grower occasionally, especially early, to assure there were no problems getting beyond the germination stage. If problems occur, there might still be time to go to another grower or to adjust the planting date.

When determining whether to accept the plant materials, look at the roots in addition to the tops. The tops and roots should be about the same in terms of density. Always remove several plants from their containers to look at the roots. The roots should extend to the bottom of the container, but they should not be root bound (wound around the inside of the container). If they are root bound, the grower did not transplant them to larger containers in a timely manner. The roots should have several well-developed rhizomes in addition to hair roots. The tops should be vigorous and as tall as the contract called for. If the tops are too short, the plants will be in danger of drowning if planted in water that is too deep. The aerenchyma should be well started in the bottom third of the aboveground biomass.

Wildlings or wild transplant collection

Wetland plants because of their tremendous root systems are readily transplanted, and the remaining plants will fill in the harvest hole rapidly. One rule of thumb is to dig no more than 1 square foot (0.09 m^2) of plant material from a 4-square-foot (0.4 m^2) area. It is not necessary to go deeper than about 5 to 6 inches (13 to 15 cm) (fig. I.D.7a–4). This will get enough of the root mass to ensure good establishment at the project site. Enough of the transplants' root system will be retained below the harvest point to allow the plants to grow back into the harvest hole in one growing season assuming good hydrology and some sediment input (Bentrup and Hoag 1998). Transplants can be taken at almost any time of the year. Collections in Idaho have been taken from March to October with little or no difference in transplant establishment success. If plugs are taken during the summer, cut the tops down to about 4 to 5 inches (10 to 13 cm) above the potential standing water height or 10 inches (26 cm), which ever is taller. Research at the Aberdeen Plant Materials Center has shown that covering the cut ends with water will not necessarily kill the plant, but significantly slows its establishment rate (except if left for longer periods) (Hoag et al. 1992). Cutting the tops also increases the survival rate of transplants that are transported long distances.

Figure I.D.7a–4 Digging wildling plug; dig no deeper than 6 inches

Generally, leaving the soil on the plug increases the establishment success by about 30 percent. Beneficial organisms typically found on the roots of the wetland plants that are important in the nitrogen and phosphorous cycles can be moved to the new site, which often will not have the organisms. However, the volume of material that needs to be transported will increase. In addition, if collections are made from a weed infested area, there is a good chance that weed seeds could be transported in the soil. Washed plugs can be inoculated with mycorrhizae purchased from dealers if the project objectives call for it. The collection location also helps determine whether the soil should be left on the plugs or washed off.

If a total of 1 cubic foot (0.09 m^2) of plant material is harvested, it is possible to get 4 to 5 individual plants plugs from the larger plug. The plugs can be either chopped with a shovel rapidly or cut relatively accurately with a small saw so they can easily fit into a predrilled, set diameter hole. To get the right length of plug, lay the large plug on its side on a sheet of plywood and use the saw to cut the bottom off level and to the desired length. After this, stand it up and cut smaller plugs off like a cake.

Make sure the length of the plug is related to the saturation zone at the planting site. The bottom of the plug needs to be in contact with the saturation zone. Match the amount of water with the wetland plant species. Ogle and Hoag (2000) display a hydrologic planting zone diagram that outlines the various hydrologic regimes. They also include a series of tables that specify which zones various species will tolerate.

Wetland transplant planting

Natural wetland systems have high species diversity. Consider the following factors when selecting plant species for the project wetland.

- Copy a nearby natural wetland.
- Identify the particular hydrology in areas where the individual plant species are growing.
- Note the water depth.
- Imagine how long the plants will be inundated.

- Determine if the plants are in flowing or relatively stagnant water. Rarely will a natural wetland be totally stagnant through time. Generally, there is water flowing into the wetland from somewhere either aboveground or from groundwater. Spring and fall overturn, as well as wind mixing, also help to circulate the water.

Next, prepare the planting area. The easiest way to plant the plugs is by flooding the planting site. Standing water is much easier to plant in than dry soil (this also ensures that the watering system, whatever it may be, works before planting is started). Make sure the soil is super saturated so that a hole can be dug by hand. This is more successful with fine soils than with coarse soils.

To transport the plugs to the planting area, place the plug trays in a Styrofoam™ cooler (you will not need the lid). Cover most of the roots with water.

At the planting site, drain off most of the water so the cooler will float. Use the cooler to move the plugs around the wetland as they are planted. Select a spot in the wetland to put a plug, reach into the water with your hand, and dig out a hole deep enough for the plug to fit. Push the plug into the hole and pack soil around it. Make sure all of the roots are covered with soil. Be careful to not dislodge the plug and expose the roots when moving around. Start at one end of the planting site and work toward the opposite end.

Spacing of the plugs is a common question. The research indicated that many wetland plants typically spread about 9 to 12 inches (23 to 30 cm) in a full growing season. In the research project, the plugs were planted on 18-inch (46-cm) centers. Even though it takes fewer plants to plant an area at a wider spacing, the research showed that plantings at wider spacing have less overall success than those planted at closer spacing. The exact reason for this is unknown, but it could be a sympathetic response to plants of the same species. If the project budget does not allow for the purchase of enough plants to cover the wetland bottom, plant the plugs on 18-inch (46 cm) centers, but plant them in copses or patches that are about 10-foot (3 m) square. Space the copses about 10 feet (3 m) apart. The copses can be planted to different species according to the hydrology. Over time, the plants will spread out into the unplanted areas.

The planting window for wetland plants is quite long. At the Aberdeen Plant Materials Center, Idaho, plugs have been planted from April through late October. Planting plugs in the fall and winter has resulted in frost heaving of the plugs so that only about a third of the plug remained in the ground. The availability of water is critical. Remember wetland plants like it hot and wet. They tend to spread faster with warmer temperatures. If planting is in the spring, it will take the plants a while to get going, but they will have a longer establishment period. Fall planting generally results in lower establishment success because of the shorter growing season and frost heaving damage.

The plants can be successfully established in a variety of soil textures. Wetland plants have been successfully established in areas that are clay with no organic matter all the way up to gravels. The biggest problem is digging the holes. The soil texture often limits the equipment available to dig the holes. A small bulldozer or tractor with a ripper tooth has been used in clay bottoms to dig lines across the bottom about 8 inches (20 cm) deep.

In general, fertilizer is not necessary. However, it really depends on the site and the soils. If during construction, the bottoms have been cut down to the subsoil and all of the naturally present nutrients have been removed, fertilization will probably be necessary unless the water coming into the wetland has a high nutrient load.

After planting, release the water into the site slowly. Young plants have not fully developed the aerenchymous material necessary for them to survive in anaerobic soils and standing water. After the initial planting, be careful not to raise the water level to more than about 1 inch (2 to 3 cm) above the substrate. Too much water at this time may stress the new plants. Maintain the water at about 1 inch (2 to 3 cm) for about 1 week to inhibit the germination and growth of any terrestrial species that may be present in the restored wetland. The water level can then be lowered to the substrate surface for 15 to 20 days. This will expose the mud surface, stimulating any wetland seeds that were brought in with the transplants to germinate as well as increase the rate of spread of the transplants. Then raise the water level 1 to 2 inches (3 to 5 cm) for another week and then lower it to the substrate surface for another 15 to 20 days. After this period, slowly raise the water level to 4 to 6 inches (10 to 15 cm) for 3 to 5 days. Continue to gradually increase the water depth to 6 to 8 inches (15 to 20 cm). The aerenchymous tissues in the plant shoots are what supply the roots with oxygen, so be careful not to raise the water over the tops of the emergent vegetation. If the plants are not showing any stress, continue to carefully raise the water level to 12 to 20 inches (30 to 50 cm) if possible.

These suggested water level depths must be modified based upon the species used. Some species will not tolerate inundation at these suggested depths or durations. When in doubt, defer to the hydrology conditions on natural reference sites where the species occurs. The goal here is to inundate the transition zone between wetland and upland as much as possible to control any invading terrestrial species. After about 20 days, lower the water level to about 2 to 3 inches (5 to 7 cm) (Hammer 1992). For the rest of the growing season, adjust the water level to maximize the desired community type.

The key to determining the appropriate water level is to monitor the emergent wetland plant community. Raise the water level if weed problems surface. Lower the water level to encourage emergent wetland plant growth and spread. The point here is to fluctuate the water level. Natural wetlands rarely have a constant water level. Many species cannot tolerate a constant water level and will begin to die out. Species more tolerant to standing water will increase. The plant diversity that was so carefully planned for will be lost.

Management during the establishment year is important to ensure that the plants do not get too much water or too little. Weed control is important especially during the establishment year because of the low water levels and exposed, unvegetated areas. A good weed control plan needs to be in place before planting. Monitoring the planting for 3 to 5 years after the establishment year helps to maintain the planting and can provide useful information for future plantings.

Recommendations

- Always match the plant species to the hydrology associated with that species.
- In general, purchase the largest plugs possible. Planting technique will often determine the size of the plugs and the ease of planting.
- Plant the plugs on 18- to 24-inch (46 to 61 cm) centers.
- Plant in patches rather than wider spacing.
- Fertilizer is generally not necessary unless the water coming into the site is relatively clean or the construction has cut into the subsoil.
- Plants tend to spread faster under saturated soil conditions than in standing water. However, terrestrial weeds will move into saturated soils much faster than flooded soils. Fluctuating the water level helps the plants spread and decreases terrestrial weed establishment.
- Water control is extremely important during the establishment year.
- Weed control needs to be planned and budgeted for at the beginning of the project.
- Monitoring is essential for the success of the project. Time and money should be allocated in the budget for this purpose and a specific person to carry it out should be identified.
- Successful wetland plantings take significant planning and a good understanding of the hydrology at each site.

References

Bentrup, G., and J.C. Hoag. 1998. The practical streambank bioengineering guide; a user's guide for natural streambank stabilization techniques in the arid and semi-arid west. USDA NRCS Plant Materials Center, Interagency Riparian/Wetland Plant Development Project, Aberdeen, ID.

Carlson, J.R. 1992. Selection, production, and use of riparian plant materials for the Western United States. Proc. Intermountain Forest Nursery Assoc., T. Landis, compiler, USDA For. Serv. Gen. Tech. Rep. RM-211, Fort Collins, CO, pp. 55-67.

Grelsson, G., and C. Nilsson. 1991. Vegetation and seedbank relationships on lakeshores. Freshwater Biol. 26:199-207.

Hammer, D.A. 1992. Creating freshwater wetlands. Lewis Publ., Boca Raton, FL, 298 pp.

Hoag, J.C. 1994. Seed collection and hydrology of six different species of wetland plants. USDA NRCS Plant Materials Center, Riparian/Wetland Project Information Series #6, Aberdeen, ID.

Hoag, J.C., and M.E. Sellers. 1995. Use of greenhouse propagated wetland plants versus live transplants to vegetate constructed or created wetlands. USDA NRCS Plant Materials Center, Riparian/Wetland Project Information Series #7, Aberdeen, ID.

Hoag, J.C., and T.A. Landis. 2001. Plant materials for riparian revegetation. Native Plant J., Vol. 2, Issue 1, January 2001

Hoag, J.C., M.E. Sellers, and M. Zierke. 1992. Interagency riparian/wetland plant development project: 4th quarter FY1992 progress report. USDA NRCS Aberdeen Plant Materials Center, Aberdeen, ID.

Hoag, J.C., M.E. Sellers, and M. Zierke. 1995. Wetland plant propagation tips. View from a wetland, No. 1 (1994-1995). USDA NRCS Plant Materials Center, Riparian/Wetland Project Newsletter, Aberdeen, ID.

Leck, M.A. 1989. Wetland seed banks. *In* Ecology of Soil Seed Banks, M.A. Leck, V.T. Packer, and R.L. Simpson (eds.), Academic Press, Inc. San Diego, CA, pp. 283-305.

Manning, M.E., S.R. Swanson, T. Svejcar, and J. Trent. 1989. Rooting characteristics of four intermountain meadow community types. J. Range Manag. 42:309–312.

Ogle, D., and J.C. Hoag. 2000. Stormwater plant materials, a resource guide. Detailed Information on Appropriate Plant Materials for Best Management Practices. City of Boise Public Works Department, Boise, ID.

Salisbury, E. 1970. The pioneer vegetation of exposed muds and its biological features. Phil. Trans. Royal Soc., London, Ser. B 259:207-255.

I.D.9 Directory of wetland plant vendors

(Janet Grabowski, NRCS Plant Materials Center, Coffeeville, Mississippi, December 2001)

Issue

Wetland mitigation, restoration, and creation projects are often hindered by a limited knowledge of wetland plant vendor locations and the materials they supply. Government, commercial, and private interests often need large quantities of selected plant species from ecological regions similar to locations of potential wetland projects. Local vendors may not necessarily supply the species of interest, or the species may not be available in the volumes required. Therefore, a current national listing of vendors is critical to support these wetland activities.

Data collection

The Directory of Wetland Plant Vendors included in this section provides a national listing of vendors indexed by scientific name of wetland plant species. It was developed by personnel at the United States Department of Agriculture, Natural Resources Conservation Service (NRCS), Jamie L. Whitten Plant Materials Center, Coffeeville, Mississippi. It is an updated version of the U.S. Army Corp of Engineers, Waterways Experiment Station's 1992 version (Wetlands Research Program Technical Report WRP–SM–1, Vicksburg, MS). A thorough search was made to identify commercial sources of wetland plants in the United States; however, it is impossible to include every potential vendor. No attempt was made to exclude wetland plant species that might have invasive properties. The inclusion of any vendor does not indicate endorsement by NRCS. The species availability information was correct based on information supplied by the vendor when entered, but NRCS does not guarantee current availability or quality of plant materials produced by any vendor.

Summary

The directory provides the following:
- A listing of wetland plant vendors and their contact information.
- A listing of obligate and facultative wetland plant species (as determined by the U.S. Fish and Wildlife Service) for which vendor sources were found.
- Vendors that supply each species.
- The propagule types (seed and/or vegetative propagule) that are available.
- A listing of alternate names (synonyms) for applicable species.

Availability of publication

A limited number of hard copies of this report will be printed and available from the Jamie L. Whitten Plant Materials Center at (662) 675-2588.

This publication may also be downloaded from the World Wide Web in PDF format at:
http://www.nhq.nrcs.usda.gov/BCS/links/links.html

Contact information

For further information or comments, please contact:

USDA, Natural Resources Conservation Service
Jamie L. Whitten Plant Materials Center
Attn: Janet Grabowski
2533 County Road 65
Coffeeville, MS 38901
(662) 675-2588
Fax (662) 675-2369
jgrabowski@ms.nrcs.usda.gov

Literature citation

Grabowski, J. 1999. Directory of wetland plant vendors in the United States. USDA-NRCS Jamie L. Whitten Plant Materials Center, Coffeeville, MS. 79 p.

Directory of Wetland Plant Vendors

Contents

Introduction

This Directory of Wetland Plant Vendors is divided into three parts.

Part1 lists the vendors of wetland plants by State. Each vendor is a assigned a Nursery Code, and these codes are listed in numerical order.

Part 2 lists only wetland plants for which vendors were found, sorted by the most currently accepted species name. This list was drawn from the species list in the Federal Register Notice 2680-2681, Volume 62, Number 12, Reed, Jr., Porter B. 1997. *Draft Revision of The National List of Plant Species That Occur in Wetlands: 1996 National Summary*, U.S. Fish and Wildlife Service (USFWS), National Wetlands Inventory, St. Petersburg, Florida, which is a proposed revision of the 1988 National Listing. Only those species in the obligate wetland (OBL) or facultative wetland (FACW, including + and –) indicator categories within some geographic region on the USFWS list were included in this directory. Wetland indicator species not listed are those for which no commercial sources were located when compiling information for this directory.

The plant nomenclature for this directory follows USDA, NRCS. 1999. *The Plants Database.* National Plant Data Center, Baton Rouge, LA, **<http://plants.usda.gov>** and was cross-checked with the PLANTS database as of May 1998.

The list contains some naturally occurring hybrid species that are designated by a genus name followed by an **X** and the species name; e.g., *Typha X glauca* (blue cattail). Those species with a **var.** (botanical variety) or **ssp.** (subspecies) designation indicate that these forms are to be used in a wetland habitat because of their increased tolerance to wet sites. For example, *Viburnum dentatum* var. *dentatum* (Southern arrow-wood) is more adapted to conditions of high soil moisture than the species, *Viburnum dentatum.* Those interested in purchasing plants for wetland use should contact the vendor to confirm the availability of the specified subspecies or variety. The common names listed are those most widely accepted for that species, although other names may be used locally. The directory includes commercially available cultivars within the listing for the plant species, but does not identify them as such.

The species are referenced by Nursery Code to the vendors listed in part 1. Codes listed for each species end in one of three letters, indicating the type of propagules available from that vendor **(p = plants or other vegetative propagules, s = seed, and b = both seed and vegetative propagules)**. Ideally, plant materials to be purchased should originate from within the same geographical region in which they will be planted. Wide differences in latitude generally have more bearing on the suitability of plant materials than similar differences in longitude; however, there are many additional factors, such as soil type, rainfall patterns, and local topography, involved in determining adaptability. Materials grown by local nurseries may not have originated from a local source, so it is highly recommended that the nursery be contacted to determine suitability of the material prior to purchase. Some of the materials listed may have been selected by horticulturists for some distinguishing feature, and would therefore not possess the genetic diversity of those obtained from a natural population. For wetland restoration and mitigation uses, it would be better to concentrate on obtaining materials from nurseries that specialize in producing wetland plants for these uses. Local NRCS field offices can assist in planning many wetland projects, and for complex plantings, consultation with a wetlands vegetation specialist is also recommended.

Part 3 lists the synonyms for the species listed in part 2. All synonyms are according to the PLANTS database. The synonyms/alternate names list will be useful for locating species that may be commonly referred to by an older name. For example, *Scirpus validus* is a former name for the currently accepted *Schoenoplectus tabernaemontani*.

To use this directory:

- Look up the species of interest. For example, plans for a wetland creation site call for *Lobelia cardinalis* (cardinal flower).
- If the species of interest cannot be located, check the alternate plant names list in part 3. If the species name is not listed in part 3, a source was not located for that plant.
- Note the Nursery Codes listed for that species that are geographically similar to the planting site and that offer the desired propagule type. For example, the site is located in Kentucky (KY) and requires plants as the propagule. Looking at the listing for cardinal flower, there are two KY vendor codes that offer plants, KY002b and KY003p.
- Go to part 1 and find the information listed for the vendor(s). For example, KY002 is the code for Shooting Star Nursery, 444 Bates Rd., Frankfort, KY 40601, (502) 223-1679.
- Contact the vendor(s) for availability and pricing information.

Directory of Wetland Plant Vendors

Part 1: Vendors

Alabama

AL001 Byers Nursery Co., Inc., P.O. Box 560, Meridianville, AL 35759
Phone: 205-828-0625; Fax: 205-859-9908

AL002 North Alabama Nursery Co., P.O. Box 67, Joppa, AL 35087
Phone: 256-586-5676; Fax: 256-586-6951

AL003 Flowerwood Nursery, Inc., 6470 Dauphin Island Pkwy., Mobile, AL 36605
Phone: 800-862-4597; Fax: 205-443-2011

AL004 Lambert Seed, P.O. Box 128, Camden, AL 36726
Phone: 334-682-4111

AL005 Great Southern Seed, P.O. Box 568, Union Springs, AL 36089
Phone: 334-738-3700

AL006 Becky's Turf Nursery, Rt. 1, Box 451-A, Tuskegee, AL 36083
Phone: 205-724-9800

AL007 Kimberly Clark, 29650 Comstock Rd., Elberta, AL 36530
Phone: 334-986-5210; Fax: 334-986-5211

Arizona

AZ001 Arizona Grain/Valley Seed Co., P.O. Box 11188, Casa Grande, AZ 85230-1188
Phone: 520-836-8713

AZ002 Mountain States Wholesale Nursery, P.O. Box 2500, Litchfield, AZ 85340-2500
Phone: 602-247-8509

AZ003 Western Sere, P.O. Box 10610, Casa Grande, AZ 85230
Phone: 520-836-8246

AZ004 Wild Seed, Inc., P.O. Box 27751, Tempe, AZ 85285
Phone: 602-276-3536; Fax: 602-276-3524

Arkansas

AR001 Weyerhaeuser Co., P.O. Box 1060, Hot Springs, AR 71902
Phone: 800-736-9330, 800-221-4898

AR002 Pittman Nursery Corp., P.O. Box 606, Magnolia, AR 71754
Phone: 800-553-6661; Fax: 870-234-6540

AR003 Kaufman Seeds, Inc., P.O. Box 398, Ashdown, AR 71822
Phone: 501-898-3328, 800-892-1082; Fax: 870-898-3302

AR004 Harmony Ridge Farm, 403 Shiloh Rd., McRae, AR 72102
Phone: 501-882-0944

AR005 Pine Ridge Gardens, 832 Sycamore Rd., London, AR 72847
Phone: 501-293-4359

California

CA001 Bamboo Sourcery, 666 Wagnon Rd., Sebastopol, CA 95472
Phone: 707-823-5866; Fax: 707-829-8106

CA002 Las Pilitas Nursery, 3232 Las Palitas Rd., Santa Margarita, CA 93453
Phone: 805-438-5992; Fax: 805-438-5993

CA003 Miniature Plant Kingdom, 4125 Harrison Grade Rd., Sabastopol, CA 95472
Phone: 707-874-2233; Fax: 707-874-3242

California (cont.)

CA004 Carter Seeds, 475 Mar Vista Dr., Vista, CA 92083
Phone: 800-872-7711, 760-724-5931; Fax: 760-724-8832

CA005 Tree of Life, P.O. Box 635, San Juan Capistrano, CA 92693
Phone: 714-728-0685; Fax: 714-728-0509

CA006 Native Sons Wholesale Nursery, Inc., 379 W. El Campo Rd., Arroyo Grande, CA 93420
Phone: 805-481-5996; Fax: 805-489-1991

CA007 Cornflower Farms, Inc., P.O. Box 896, Elk Grove, CA 95759
Phone: 916-689-1015; Fax: 916-689-1968

CA008 Forest Seeds of California, 1100 Indian Hill Rd., Placerville, CA 95667
Phone: 916-621-1551; Fax: 916-621-1040

CA009 North Coast Native Nursery, P.O. Box 744, Petaluma, CA 94953
Phone: 707-769-1213; Fax: 707-769-1230

CA010 S & S Seeds, P.O. Box 1275, Carpinteria, CA 93014
Phone: 805-684-0436; Fax: 805-684-2798

CA011 Clyde Robin Seed Co., 3670 Enterprise Ave., Hayward, CA 94545
Phone: 510-785-0425, 800-647-6475; Fax: 510-785-6463

CA012 Menzies' Native Nursery, P.O. Box 9, 10805 N. Old Stage Rd., Weed, CA 96094-0009
Phone: 916-938-4858; Fax: 916-938-4777

CA013 Elkhorn Native Plant Nursery, Box 270, Moss Landing, CA 95039
Phone: 831-763-1207; Fax: 831-763-1659

CA014 Stover Seed Company, P.O. Box 21488, Los Angeles, CA 90021
Phone: 213-626-9668

CA015 Peaceful Valley Farm Supply, P.O. Box 2209, Grass Valley, CA 95945
Phone: 530-272-4769

CA016 Kamprath Seed Co. LLC, 205 Stockton St., Manteca, CA 95337
Phone: 800-466-9959

CA017 Freshwater Farms, Inc., 5851 Myrtle Ave., Eureka, CA 95503-9510
Phone: 800-200-8969, 707-444-8261; Fax: 707-442-2490

Colorado

CO001 Country Lane Wholesale Nursery, 2979 N. Hwy 83, Franktown, CO 80116
Phone: 303-688-2442, 800-375-8696; Fax: 303-688-5978

CO002 Dean Swift Seed Co., P.O. Box B, Jaroso, CO 81138
Phone: 719-672-3739; Fax: 719-672-3865

CO003 Little Valley Nurseries, Inc., 13022 E. 136th Ave., Brighton, CO 80601
Phone: 303-659-6708, 800-221-3241; Fax: 303-659-6886

CO004 Aquatic and Wetland Co., 9999 Weld County Rd., Fort Lupton, CO 80621
Phone: 303-442-4766; Fax: 303-857-2455

CO005 Green Acres Nursery, 4990 McIntyre St., Golden, CO 80403
Phone: 303-279-8204, 888-279-8204; Fax: 303-278-1832

Colorado (cont.)

CO006 Applewood Seed Co., 5310 Vivian St., Arvada, CO 80002
Phone: 303-431-7333; Fax: 303-467-7886

CO007 Arkansas Valley Seeds, Inc., P.O. Box 270, Rocky Ford, CO 81067
Phone: 719-254-7469

CO008 Colorado State Forest Service, Colorado State Univ.,
Foothills Campus, Bldg. 1061, Ft. Collins, CO 80523
Phone: 303-491-8429

CO009 Sharp Bros. Seed Co., 101 E. 4th St. Rd., Greely, CO 80631
Phone: 970-356-4710; Fax: 970-356-1267

Connecticut

CT001 Imperial Nurseries, P.O. Box 120, Granby, CT 06035
Phone: 800-343-3132; Fax: 860-653-2919

CT002 Sunny Border Nurseries, Inc., 1709 Kensington Rd.,
Kensington, CT 06037
Phone: 860-828-0321, 800-732-1627; Fax: 860-828-9318

CT003 Casterano's Greenhouses and Farms, Inc., 1030 S. Meriden Rd.,
Chesire, CT 06410
Phone: 203-272-6444, 203-272-4563; Fax: 203-271-0496

CT004 Beardsley Gardens, 157 Gay St., Rt. 41, Sharon, CT 06069
Phone: 860-364-0727

CT005 Broken Arrow Nursery, 13 Broken Arrow Rd., Hamden, CT 06518
Phone: 203-288-1026; Fax: 203-287-1035

CT006 Hop River Nursery, 251 Hop River Rd., Route 6, Bolton, CT 06043
Phone: 860-646-7099; Fax: 860-646-7099

CT007 Logee's Greenhouses, 141 North St., Danielson, CT 06239-1939
Phone: 888-330-8038

CT008 Shepherd's Garden Seeds, 30 Irene St., Torrington, CT 06790-6658
Phone: 860-482-3638; Fax: 860-482-0532

CT009 New England Environmental Services, Blackledge River Nursery,
155 Jerry Daniels Rd., Marlborough, CT 06447
Phone: 860-295-1022

Delaware

DE001 Joseph Wick Nurseries, LTD., 3902 Brenford Rd., Smyrna, DE 19977
Phone: 302-653-9000, 800-722-WICK; Fax: 302-653-0746

DE002 Forest View Nursery, Inc, 1313 Blackbird Forest Rd.,
Clayton, DE 19938
Phone: 302-653-7757, 302-653-9165; Fax: 302-653-TREE

Florida

FL001 Central Florida Lands & Timber, Inc., Rt. 1, Box 899,
Mayo, FL 32066
Phone: 904-294-1211; Fax: 904-294-3416

FL002 Florida Keys Native Nursery, Inc., 102 Mohawk St.,
Tavernier, FL 33070
Phone: 305-852-2636, 305-852-5515

FL003 Indian Trails Nursery, 6315 Park Lane, Lake Worth, FL 33467
Phone: 561-641-9488; Fax: 561-641-9309

Florida (cont.)

FL004 Native Tree Nursery, Inc., 17250 S.W. 232nd St., Homestead, FL 33170
Phone: 305-247-4499; Fax: 305-247-4502

FL005 The Liner Distributor, Inc., P.O. Box 1389, Homestead, FL 33090
Phone: 305-247-5568, 800-330-5568; Fax: 305-248-0710

FL006 Pine Breeze Nursery, P.O. Box 702, Bokeelia, FL 33922
Phone: 941-283-7200

FL007 Sunco, 2269 Second Ave. North, Lake Worth, FL 33461
Phone: 407-586-7402; Fax: 561-588-9486

FL008 Native Plant Nursery, 3333 Sanibel Captiva Rd., Sanibel, FL 33957
Phone: 941-472-1932; Fax: 941-472-6421

FL009 Urban Forestry Services, Rt. 2, Box 940, Micanopy, FL 32667
Phone: 352-466-3919; Fax: 352-466-3280

FL010 Okefenokee Growers, P.O. Box 4488, Jacksonville, FL 32201
Phone: 904-356-4881, 800-356-4881; Fax: 904-356-4884

FL011 Coastal & Native Plant Specialties, Inc., 5951 Oglesby Rd., Milton, FL 32570
Phone: 850-623-6287, 888-884-2500; Fax: 850-626-2684

FL012 San Felasco Nurseries, Inc., 7315 NW 126 St., Gainesville, FL 32653
Phone: 352-332-1220, 800-933-9638; Fax: 352-332-3113

FL013 Mandarin Native Plants, 13500 Mandarin Rd., Jacksonville, FL 32223
Phone: 904-268-2904, 904-384-3065

FL014 Superior Trees, Inc., P.O. Box 9325, Lee, FL 32059
Phone: 904-971-5159

FL015 Environmental Equities, Inc., P.O. Box 7180, Hudson, FL 34674
Phone: 813-856-1519, 941-355-1267

FL016 The Liner Farm, Inc., P.O. Box 701369, St. Cloud, FL 34770
Phone: 407-892-1484, 800-330-1484; Fax: 407-892-3593

FL017 Aquatic Plants of Florida, Inc., , Myakka City, FL 33551
Phone: 941-952-9886, 800-266-1272; Fax: 941-952-0474

Georgia

GA001 Van Bloem Gardens, Eastern Division, 1295 Bluegrass Lakes Parkway, Alpharetta, GA 30004
Phone: 770-667-3344, 800-683-2563; Fax: 770-751-0708

GA002 Spandle Nurseries, Rt. 2, Box 125, Claxton, GA 30417
Phone: 800-553-5771

GA003 Adams-Briscoe Seed Co., P.O. Box 19, Jackson, GA 30233-0019
Phone: 770-775-7826; Fax: 770-775-7122

GA004 Mark Latimore, School of Agriculture, Fort Valley State College, Ft. Valley, GA 31030

GA005 Pennington Seed, P.O. Box 290, Madison, GA 30650
Phone: 800-277-1412

Idaho

ID001 Silver Springs Nursery, HC 62, Box 86, Moyie Springs, ID 83845
Phone: 208-267-5753; Fax: 208-267-5753

ID002 Northplan Seed Products, P.O. Box 9107, Moscow, ID 83843
Phone: 208-882-8040; Fax: 208-882-7446

ID003 Clifty View Nursery, Rt. 1, Box 509, Bonners Ferry, ID 83805
Phone: 208-267-7129; Fax: 208-267-8559

ID004 High Altitude Gardens, P.O. Box 1048, Hailey, ID 83333
Phone: 208-788-4363; Fax: 208-788-3452

ID005 Jacklin Seed Company, West 5300 River Bend Ave.,
Post Falls, ID 83854
Phone: 208-773-7581

ID006 Globe Seed and Feed Company, 224 4th Ave. South,
Twin Falls, ID 83301
Phone: 208-733-1373

ID007 Clayton Tree Farm, 6622 Joplin Rd., Nampa, ID 83687-8112
Phone: 208-286-7801; Fax: 208-286-7802

ID008 Reggear Tree Farms, 1525 Loseth Rd., Orofino, ID 83544
Phone: 208-476-7739, 208-476-7364; Fax: 208-476-7429

Illinois

IL001 Midwest Wildflowers, Box 64, Rockton, IL 61072
Phone: 815-624-7040

IL002 V & J Seed Farms, Inc., P.O. Box 82, Woodstock, IL 60098
Phone: 815-338-4029; Fax: 815-338-4029

IL003 Owen Nursery, 2300 E. Lincoln St., Bloomington, IL 61701
Phone: 309-663-9551

IL004 The Natural Garden, Inc., 38W443 Hwy 64, St. Charles, IL 60175
Phone: 630-584-0595; Fax: 630-584-0432

IL005 Burgess Seed & Plant Co., 905 Four Seasons Rd.,
Bloomington, IL 61701
Phone: 309-663-9551

IL006 Four Seasons Nursery, Division of Plantron, Inc., 1706 Morrissey
Dr., Bloomington, IL 61704
Phone: 309-663-9551

Indiana

IN001 C.M. Hobbs & Sons, Inc., P.O. Box 31227, Indianapolis, IN 46231
Phone: 317-247-4478, 800-428-6765; Fax: 317-241-9253

IN002 Maschmeyer's Nursery, Inc., 3009 E. 500 North, P.O. Box 8,
Whiteland, IN 46184
Phone: 317-535-7541; Fax: 317-535-9403

IN003 Spence Restoration Nursery, P.O. Box 546, 2220 E. Fuson Rd.,
Muncie, IN 47308
Phone: 765-286-7154; Fax: 765-286-0264

Iowa

IA001 Cascade Forestry Nursery, 22033 Fillmore Rd., Cascade, IA 52033
Phone: 319-852-3042; Fax: 319-852-5004

IA002 Mount Arbor Nurseries, 400 N. Center, P.O. Box 129,
Shenandoah, IA 51601
Phone: 800-831-4125; Fax: 712-246-1841

IA003 Osenbaugh Grass Seeds, RR 1, Box 44, Lucas, IA 50151
Phone: 800-582-2788; Fax: 515-766-6795

IA004 Henry Field's Seed & Nursery Co., 415 N. Burnett,
Shenandoah, IA 51602
Phone: 605-665-9391, 605-665-4491; Fax: 605-665-2601

IA005 Ion Exchange, 1878 Old Mission Dr., Harpers Ferry, IA 52146
Phone: 319-535-7231; Fax: 319-535-7362

Kansas

KS001 Sharp Bros. Seed Co., Box 140, Healy, KS 67850
Phone: 316-398-2231

KS002 Mott Ranch, Rt. 1, Box 79, Iuka, KS 67006
Phone: 316-546-2575; Fax: 316-546-2250

KS003 Valley Feed and Seed, 1903 S. Meridian, Wichita, KS 67213
Phone: 316-942-2278; Fax: 316-942-2268

KS004 Glen Snell, 300 N. Adams, Medicine Lodge, KS 67104
Phone: 316-886-5075; Fax: 316-886-3008

Kentucky

KY001 Valley Hill Nurseries, 4251 Bloomfield Rd., Springfield, KY 40069
Phone: 606-336-9017; Fax: 606-336-0470

KY002 Shooting Star Nursery, 444 Bates Rd., Frankfort, KY 40601
Phone: 502-223-1679

KY003 Dabney Herbs, P.O. Box 22061, Louisville, KY 40252
Phone: 502-893-5198; Fax: 502-893-5198

Louisiana

LA001 Holloway's Nursery, Inc., 11528 Hwy 165 S., P.O. Box 339,
Forest Hill, LA 71430
Phone: 318-748-6803, 800-634-2815; Fax: 318-748-7204

LA002 Native Nurseries, 320 N. Theard St., Covington, LA 70433
Phone: 504-892-5424

LA003 Doug Young's Nursery, P.O. Box 39, Forest Hill, LA 71430
Phone: 318-748-8207, 318-748-6071; Fax: 318-748-8208

LA004 The Bosch Nursery, Inc., 18874 Hwy 4, Jonesboro, LA 71251
Phone: 318-259-9484

LA005 Louisiana Dept. of Agriculture and Forestry, P.O. Box 1628,
Baton Rouge, LA 70821
Phone: 504-925-4515

LA006 Don Huemann Greenhouse and Lab, 808 Rue Chartres,
Metarie, LA 70005
Phone: 504-833-2473

LA007 Live Oak Gardens, Ltd., 10106 Jefferson Island Rd.,
New Iberia, LA 70560
Phone: 800-725-5625, 318-367-3485; Fax: 318-364-1605

Louisiana (cont.)

LA008 Louisiana Forest Seed Co., Inc., 303 Forestry Rd.,
Lecompte, LA 71346
Phone: 318-443-5026; Fax: 318-487-0316

LA009 Coastal Plants, Inc., 2320 W. Alcide Dr., Abbeville, LA 70510
Phone: 318-898-3098

Maine

ME001 Johnny's Selected Seeds, 1 Foss Hill Rd., RR 1 Box 2580,
Albion, ME 04910
Phone: 207-437-9294, 207-437-4301; Fax: 800-437-4290

ME002 Daystar, 1270 Hallowell-Litchfield Rd., West Gardiner, ME 04345
Phone: 207-724-3369

ME003 Pinetree Garden Seeds, Box 300, New Gloucester, ME 04260
Phone: 207-926-3400; Fax: 888-527-3337

Maryland

MD001 Lilypons Water Gardens, 6800 Lilypons Rd., P.O. Box 10,
Buckeystown, MD 21717
Phone: 301-874-5133, 800-999-5459; Fax: 800-879-5459

MD002 Environmental Concern, Inc., 210 W. Chew Ave., P.O. Box P,
St. Michaels, MD 21663
Phone: 410-745-9620; Fax: 410-745-3517

MD003 Chesapeake Nurseries, Inc., 27571 Pemberton Dr.,
Salisbury, MD 21801
Phone: 800-772-1118

Massachusetts

MA001 F.W. Schumacher Co., Inc., 36 Spring Hill Rd., Sandwich, MA 02563
Phone: 508-888-0659; Fax: 508-833-0322

MA002 Bestman Green Systems, 53 Mason St., Salem, MA 01970
Phone: 978-741-1166; Fax: 978-741-3780

MA003 New England Wetland Plants, Inc., 800 Main St.,
Amherst, MA 01002
Phone: 413-256-1752; Fax: 413-256-1092

MA004 Tripple Brook Farm, 37 Middle Rd., Southampton, MA 01073
Phone: 413-527-4626; Fax: 413-527-9853

Michigan

MI001 Walters Gardens, Inc., P.O. Box 137, Zeeland, MI 49464
Phone: 616-772-4697, 888-925-8377; Fax: 800-752-1879

MI002 Zelenka Nursery, Inc., 16127 Winans St., Grand Haven, MI 49417
Phone: 616-842-1367, 800-253-3743; Fax: 800-842-0321

MI003 Vans Pines, Inc., 7550 144th Ave., West Olive, MI 49460
Phone: 616-399-1620, 800-888-7337; Fax: 616-399-1652

MI004 Cottage Gardens, Inc., Michigan Division, 2611 S. Waverly Hwy,
Lansing, MI 48911-6399
Phone: 800-523-1923, 517-882-5728; Fax: 517-882-4290

MI005 The Garden Store, 324 Meadow Creek Lane,
Grand Rapids, MI 49550-1000
Phone: 800-582-8649, 616-735-2130; Fax: 800-496-2852

Minnesota

MN001 Orchid Gardens, 2232 139th Ave. N.W., Andover, MN 55304
Phone: 612-755-0205

MN002 Bailey Nurseries, Inc., 1325 Bailey Rd., St. Paul, MN 55119
Phone: 612-459-9744; Fax: 612-459-5100

MN003 Cross Nurseries, Inc., 19774 Kenwood Trail West,
Lakeville, MN 55044
Phone: 612-469-2414, 888-217-0826; Fax: 612-469-1844

MN004 Prairie Restorations, Inc., P.O. Box 327, Princeton, MN 55371
Phone: 612-389-4342

MN005 Hartman Tree Farm, 8099 Bavaria Rd., Victoria, MN 55386
Phone: 612-443-2990, 800-473-4812; Fax: 612-443-2835

MN006 Bachman's Nursery Wholesale Center, 6877 235th St. West,
Farmington, MN 55024
Phone: 612-463-3288, 800-525-6641; Fax: 612-463-4747

MN007 Mohn Frontier Seed Co., RR1, Box 152, Cottonwood, MN 56299
Phone: 507-423-6482; Fax: 507-423-5552

MN008 Holly Lane Iris Gardens/Worel's, 10930 Holly Lane North,
Osseo, MN 55369
Phone: 612-420-4876

MN009 Prairie Moon Nursery, Rt. 3, Box 163, Winona, MN 55987
Phone: 507-452-1362; Fax: 507-454-5238

MN010 Busse Gardens, 5873 Oliver Ave. S.W., Cokato, MN 55321
Phone: 320-286-2654, 800-544-3192; Fax: 320-286-6601

MN011 Feder's Prairie Seed Co., 12871 380th Ave., Blue Earth, MN 56013
Phone: 507-526-3049; Fax: 507-526-3509

MN012 Law's Nursery, Inc., 13030 Maycrest Ave. Court South,
Hastings, MN 55033
Phone: 612-437-9119; Fax: 612-438-3097

MN013 Shooting Star Native Seeds, Hwy 44 & CR 33, P.O. Box 648,
Spring Grove, MN 55974
Phone: 507-498-3944; Fax: 507-498-3953

MN014 Farmer Seed & Nursery, 818 NW 4th St., Faribault, MN 55021
Phone: 507-334-1623

MN015 Premium Seed Co., 7800 E. Hwy 101, Shakopee, MN 55379
Phone: 612-496-1783

MN016 Norfarm Seeds, Inc., P.O. Box 725, Bemidji, MN 56619
Phone: 218-751-3350; Fax: 218-751-0485

MN017 TEC, P.O. Box 539, Osseo, MN 55369-0539

MN018 Peterson Seed Co., Box 346, Savage, MN 55378
Phone: 612-445-2606

MN019 Albert Lea Seedhouse, 1414 W. Main, P.O. Box 127,
Albert Lea, MN 56007
Phone: 501-373-3161

MN020 Kelly Nurseries, Division of Plantron, Inc., 410 8th Ave. NW,
Faribault, MN 55021
Phone: 507-334-1623

Mississippi

MS001 Delta View Nursery, P.O. Box 157, Scott, MS 38772
Phone: 800-511-7333; Fax: 601-742-3472

MS002 Green Forest Nursery, 1478 Old Hwy 26, Perkinston, MS 39573
Phone: 601-928-7266; Fax: 601-928-5008

MS003 Jarrell's Aquatic Nursery, 470 Pine Grove Rd., Picayune, MS 39466
Phone: 601-798-1720

MS004 Weyerhauser, P.O. Box 2288, Columbus, MS 39704
Phone: 800-635-0162

MS005 Bear Creek Nursery, 1267 Patrick Rd., Canton, MS 39046
Phone: 601-898-8071; Fax: 601-605-1001

MS006 Bulldog Nursery, 1738 McIngvale, Hernando, MS 38632
Phone: 601-429-6048

MS007 Meadowview Nursery, 449 F.Z. Goss Rd., Picayune, MS 39466
Phone: 601-799-0088

MS008 Raintree Center Aquatic Gardens, 119 E. 2nd St.,
Pass Christian, MS 39571
Phone: 228-452-3137

MS009 Pine Belt Mental Health, 1723 Stanley St., Hattiesburg, MS 39401

Missouri

MO001 Sharp Bros. Seed Co., 396 S.W. Davis St., Clinton, MO 64735
Phone: 816-885-7551, 800-451-3779; Fax: 816-885-8647

MO002 Forrest Keeling Nursery, P.O. Box 135, Elsberry, MO 63343
Phone: 573-898-5571; Fax: 573-898-5803

MO003 Hamilton Seeds, 16786 Brown Rd., Elk Creek, MO 65464
Phone: 417-967-2190

MO004 Stark Brothers Nurseries, Louisiana, MO 63353
Phone: 800-325-4180

MO005 Heartland Nursery Co., 311 Main St., New Madrid, MO 63869-1942
Phone: 573-748-5515; Fax: 573-748-9155

MO006 J & J Seed Co., 29341 210 St., Gallatin, MO 64640
Phone: 660-663-3165; Fax: 660-663-2301

MO007 Royal Seed, 225 Florence Rd., St. Joseph, MO 64504
Phone: 800-753-0990; Fax: 816-238-7849

MO008 Flick Seed Co., 1781 NW 50th Rd., Kingsville, MO 64061
Phone: 816-597-3822; Fax: 816-597-3663

MO009 Missouri Wildflower Nursery, 9814 Pleasant Hill,
Jefferson City, MO 65109
Phone: 573-496-3492; Fax: 573-496-3003

MO010 Shepard's Farms, Rt. 1, Box 7, Clifton Hills, MO 65244
Phone: 816-261-4567; Fax: 816-261-4422

MO011 Mangelsdorf Seed Co., 1415 N. 13th St., St. Louis, MO 63106-4424
Phone: 314-421-1415, 800-467-7333; Fax: 314-421-1954

Montana

MT001 Bitterroot Native Growers, Inc., 445 Quast Lane,
Corvallis, MT 59828
Phone: 406-961-4991; Fax: 406-961-4626

MT002 Blake Nursery, Otter Creek Rd., HC 87 Box 2240,
Big Timber, MT 59011
Phone: 406-932-4195

MT003 Westland Seed, Inc., Box 57, Charlo, MT 59824
Phone: 406-644-2202, 800-547-3335; Fax: 406-676-4101

MT004 Big Sky Wholesale Seeds, Inc., Box 852, Shelby, MT 58474
Phone: 406-434-5011; Fax: 406-434-5014

MT005 Cashman Nursery, P.O. Box 242, Bozeman, MT 59715
Phone: 406-587-3406

MT006 Valley Nursery, Box 4845, Helena, MT 59604

MT007 Lawyer Nursery, Inc., 950 Hwy 200 West, Plains, MT 59859-9706
Phone: 406-826-3881, 800-551-9875; Fax: 406-826-5700

MT008 Montana Seeds, Inc., Rt. 3, Conrad, MT 59424
Phone: 406-278-5547

MT009 United AgriProducts, 1400 Minnesota, Billings, MT 59101
Phone: 406-252-8012

MT010 Union Seed Co., 418 Albert St., Billings, MT 59101-3301
Phone: 406-252-0568

MT011 Bill Skorupa, Box 1211, Bridger, MT 59014
Phone: 406-662-3358

MT012 Treasure State Seed Co., Box 698, Fairfield, MT 59436
Phone: 406-467-2557

MT013 Valley Feed, 1370 Hwy 10 West, Livingston, MT 59047
Phone: 406-222-1132

MT014 Circle S Seeds of Montana, Inc., Box 130, Three Forks, MT 59752
Phone: 406-285-3269

MT015 Bitterroot Nursery, 521 Eastside Hwy, Hamilton, MT 59840
Phone: 406-961-3806; Fax: 406-961-3765

Nebraska

NE001 Gamagrass Seed Co., Rt. 1, Box 114A, Falls City, NE 68355
Phone: 402-245-5928, 800-367-2879

NE002 Stock Seed Farms, 28008 Mill Rd., Murdock, NE 68407
Phone: 402-867-3771, 800-759-1520; Fax: 402-867-2442

NE003 The Fragrant Path, P.O. Box 328, Fort Calhoun, NE 68023

NE004 Miller Grass Seed Co., P.O. Box 81823, Lincoln, NE 68501-1823

NE005 Arrow Seed Co., 126 N. 10th, Broken Bow, NE 68822
Phone: 308-872-6826; Fax: 308-872-6945

NE006 Heritage Seed Co., Inc., P.O. Box 544, Crawford, NE 69339
Phone: 308-665-1672; Fax: 308-665-1526

NE007 De Giorgi Seed Co., 6011 N St., Omaha, NE 68117
Phone: 402-731-3901; Fax: 402-731-8475

NE008 Laux Seed Farm, Inc., HC 85, P.O. Box 48, Bridgeport, NE 69336
Phone: 308-262-0512

Nebraska (cont.)

NE009 Osler Seed Farms, HC 55, P.O. Box 123, Elsie, NE 69134
Phone: 308-228-2287

NE010 Cenex Land O' Lakes, 1431 S. Webb Rd., Grand Island, NE 68803
Phone: 308-384-1111

NE011 Bluebird Nursery, Inc., P.O. Box 460, Clarkson, NE 68629
Phone: 402-892-3457, 800-356-9164; Fax: 402-892-3738

Nevada

NV001 Nevada Division of Forestry, Washoe Nursery, 885 Eastlake Blvd., Carson City, NV 89704
Phone: 702-849-0213; Fax: 702-849-2058

New Hampshire

NH001 Lowe's Own-Root Roses, 6 Sheffield Rd., Nashua, NH 03062
Phone: 603-888-2214, 704-859-2571

NH002 Tree Dimensions, RR 1, Box 216A, North Haverhill, NH 03774
Phone: 603-787-6825

New Jersey

NJ001 Wild Earth Native Plant Nursery, P.O. Box 7258, Freehold, NJ 07728
Phone: 732-308-9777; Fax: 732-308-9777

NJ002 Croshaw Nursery, P.O. Box 339, Columbus, NJ 08022
Phone: 609-298-0477; Fax: 609-298-6388

NJ003 Jonathan Green Seeds, P.O. Box 326, Squankum-Yellowbrook Rd., Farmingdale, NJ 07727
Phone: 800-243-0047, 800-526-2303; Fax: 732-938-5788

NJ004 Princeton Nurseries, P.O. Box 185, Allentown, NJ 08501
Phone: 800-916-1776

NJ005 Arrowwood Nursery, Inc., 870 W. Malaga Rd., Williamstown, NJ 08094
Phone: 609-697-6045, 609-875-4889; Fax: 609-697-6050

NJ006 Visconti Nursery, 1459 Centerton Rd., Pittsgrove, NJ 08318
Phone: 906-358-6644

NJ007 Thompson & Morgan, Inc., P.O. Box 1308, Jackson, NJ 08527-0308
Phone: 800-274-7333; Fax: 888-466-4769

NJ008 Pinelands Nursery, 323 Island Rd., Columbus, NJ 08022
Phone: 609-291-9486, 800-667-2729; Fax: 609-298-8939

NJ009 Coastal Natives Nursery, Inc., P.O. Box 42, Mauricetown, NJ 08329
Phone: 609-785-1102; Fax: 609-785-9301

NJ010 Winslow Conservancy, 303 Messina Ave., Hammonton, NJ 08037
Phone: 609-561-0628

VA004 Pinelands Nursery, 323 Island Rd., Columbus, NJ 08022
Phone: 609-291-9486, 800-667-2729; Fax: 609-298-8939

New Mexico

NM001 Curtis & Curtis, Inc., Star Rt., Box 8A, Clovis, NM 88101
Phone: 505-762-4759; Fax: 505-763-4213

New York

NY001 Peter Pauls Nurseries, 4665 Chapin Rd., Canadaigua, NY 14424
Phone: 716-394-7397; Fax: 716-394-4122

NY002 Plantage, Inc., P.O. Box 28, Cutchogue, NY 11935
Phone: 516-734-6832; Fax: 516-734-7550

NY003 Baier Lustgarten Farms & Nurseries, 1130 Middle Country Rd., Middle Island, NY 11953-2527
Phone: 516-924-3444; Fax: 516-924-2211

NY004 Sheffield's Tree & Shrub Seed, 273 Auburn Rd., Rt. 34, Locke, NY 13092
Phone: 315-497-1058; Fax: 315-497-1059

NY005 Roslyn Nursery, 211 Burrs Lane, Dix Hills, NY 11746
Phone: 516-643-9347; Fax: 516-427-0894

NY006 Congdon & Weller Wholesale, Inc., P.O. Box 1507, Mile Block Rd., North Collins, NY 14111
Phone: 800-345-8305; Fax: 716-337-0203

NY007 Seedway, Box 250, Hall, NY 14463
Phone: 716-526-6391

North Carolina

NC001 N.C. Division Forest Resources, P.O. Box 29581, Raleigh, NC 27626
Phone: 919-733-2162, 888-NCTREES; Fax: 704-438-6002

NC002 McLamb Nursery, Inc, 640 Greenleaf Rd., Angier, NC 27501
Phone: 919-894-3709; Fax: 919-894-2446

NC003 Perry's Water Gardens, 1831 Leatherman Gap Rd., Franklin, NC 28734
Phone: 704-524-3264; Fax: 704-369-2050

NC004 Niche Gardens, 1111 Dawson Rd., Chapel Hill, NC 27516
Phone: 919-967-0078; Fax: 919-967-4026

NC005 We-Du Nurseries, Rt. 5, Box 724, Marion, NC 28752
Phone: 704-738-8300; Fax: 704-738-8131

NC006 Boothe Hill Wildflower Seed, 921 Boothe Hill, Chapel Hill, NC 27514
Phone: 919-967-4091

NC007 Camellia Forest Nursery, 125 Carolina Forest Rd., Chapel Hill, NC 27516
Phone: 919-967-5529, 919-968-0504

NC008 Hoffman Nursery, 5520 Bahama Rd., Rougemont, NC 27572
Phone: 919-479-6620, 800-203-8590; Fax: 919-471-3100

NC009 H. Burkert & Co., 37 Covil Ave., Wilmington, NC 28403
Phone: 910-763-4600

North Dakota

ND001 Heartland, Inc., Box 1877, Bismarck, ND 58502
Phone: 701-223-4065

ND002 Chesak Seedhouse, 220 N. 23rd St., Bismarck, ND 58501
Phone: 701-223-0391

Ohio

OH001 Girard Nurseries, P.O. Box 428, Geneva, OH 44041
Phone: 440-466-2881; Fax: 440-466-3999

OH002 Bluestone Perennials, 7211 Middle Ridge Rd., Madison, OH 44057
Phone: 440-428-7535, 800-852-5243; Fax: 440-428-7198

OH003 Lake County Nursery, Inc., Rt. 84, P.O. Box 122, Perry, OH 44081
Phone: 800-522-5253; Fax: 800-699-3114

OH004 Manbeck Nurseries, Inc., P.O. Box 309, New Knoxville, OH 45871
Phone: 419-753-2488; Fax: 419-753-2712

OH005 G.S. Grimes Seeds, 11335 Concord-Hambden, Concord, OH 44077
Phone: 800-241-7333; Fax: 216-352-1800

OH006 Sunnybrook Farms, P.O. Box 6, Chesterland, OH 44026
Phone: 216-729-7232

OH007 Springbrook Gardens, Inc., 6776 Heisley Rd., Mentor, OH 44061
Phone: 216-255-3059; Fax: 216-255-9535

OH008 Klyn Nurseries, Inc., 3322 S. Ridge Rd., P.O. Box 343,
Perry, OH 44081
Phone: 440-259-3811, 800-860-8104; Fax: 440-259-3338

OH009 The Cottage Gardens, Inc., Ohio Division, 4992 Middle Ridge Rd.,
Perry, OH 44081
Phone: 877-377-5877, 440-259-2900; Fax: 440-259-3154

OH010 Spring Hill Nurseries, 110 W. Elm St., Tipp City, OH 45371
Phone: 800-582-8527; Fax: 800-991-2852

Oklahoma

OK001 Greenleaf Nursery Co., HC-72, Box 163, Park Hill, OK 74451
Phone: 800-331-2982, 800-237-3147

OK002 Johnston's, P.O. Box 1392, Enid, OK 73702
Phone: 800-375-4613; Fax: 580-249-5324

OK003 Grasslander, Rt. 1, Box 56, Hennessey, OK 73742
Phone: 405-853-2607

OK004 Tri-B Nursery, Inc., P.O. Box 436, Tahlequah, OK 74465
Phone: 800-244-6157, 918-772-3428; Fax: 918-772-2607

OK005 Park Hill Plants & Trees, Inc., P.O. Box 368, Park Hill, OK 74451
Phone: 918-456-4548; Fax: 918-456-5900

OK006 B.G. Barby Ranch, P.O. Box 1660, Woodward, OK 73802

OK007 Oklahoma Forestry Services, 2800 N. Lincoln Blvd.,
Oklahoma City, OK 73105-4298
Phone: 405-521-3864

Oregon

OR001 Meyer Nursery & Orchards, 3795 Gibson Rd., Salem, OR 97304
Phone: 503-364-3076, 800-779-0440; Fax: 503-364-3407

OR002 The Bovees Nursery, 1737 S.W. Coronado, Portland, OR 97219
Phone: 503-244-9341, 800-435-9250

OR003 John Holmlund Nursery Co., 29285 S.E. Hwy 212, Boring, OR 97009
Phone: 503-663-6650, 800-643-6650; Fax: 503-663-2356

OR004 Russell Graham Nursery, 4030 Eagle Crest Rd. N.W.,
Salem, OR 97304
Phone: 503-362-1135

Oregon (cont.)

OR005 Carlton Plants, 14301 S.E. Wallace Rd., P.O. Box 398,
Dayton, OR 97114
Phone: 503-868-7971, 800-398-8733; Fax: 800-442-1452

OR006 Iseli Nursery, Inc., 30590 S.E. Kelso Rd., Boring, OR 97009
Phone: 503-663-3822, 800-777-6202; Fax: 503-663-0202

OR007 T.H. Belcher Nursery, Inc., 33755 S.E. Bluff Rd., Boring, OR 97009
Phone: 503-663-3593; Fax: 503-663-0619

OR008 Cascadian Nurseries, Inc., 13495 N.W. Thompson Rd.,
Portland, OR 97229
Phone: 503-645-3350; Fax: 503-645-0333

OR009 Harold M. Miller Landscape Nursery, P.O. Box 989,
Jefferson, OR 97352
Phone: 503-399-1599; Fax: 503-364-7552

OR010 Bell Maple Nursery, 33901 S.E. Bluff Rd., Boring, OR 97009
Phone: 503-663-5780; Fax: 503-663-5782

OR011 Greer Gardens, 1280 Goodpasture Island Rd., Eugene, OR 97401
Phone: 541-686-8266, 800-548-0111; Fax: 541-686-0910

OR012 Jackson & Perkins, 1 Rose Lane, Medford, OR 97501
Phone: 800-292-4769, 800-872-7673; Fax: 800-242-0329

OR013 D. Wells Nursery, P.O. Box 336, Hubbard, OR 97032
Phone: 503-982-1012; Fax: 503-981-8420

OR014 Forest Farm, 990 Tetherow Rd., Williams, OR 97544-9599
Phone: 541-846-7269; Fax: 541-846-6963

OR015 Lew's Lakeshore Nursery, 581 Lancaster Dr. SE, Suite 275,
Salem, OR 97301-5642
Phone: 541-504-4569; Fax: 541-504-8349

OR016 Heritage Seedlings, Inc., 4199 75th Ave. SE, Salem, OR 97301-9242
Phone: 503-585-9835; Fax: 503-371-9688, 800-727-8744

Pennsylvania

PA001 Appalachian Gardens, Box 87, Waynesboro, PA 17268
Phone: 717-762-4312, 888-327-5483

PA002 W. Atlee Burpee & Co., 300 Park Ave., Warminster, PA 18974
Phone: 800-333-5808, 800-888-1447; Fax: 800-487-5530

PA003 Eisler Nurseries, Rt. 422, Box 465, Prospect, PA 16052
Phone: 412-865-2830; Fax: 412-865-9018

PA004 Ecoscience, Inc., R.R. 4, Box 4294, Moscow, PA 18444
Phone: 717-842-7631, 888-4-WETLANDS; Fax: 717-842-9976

PA005 Musser Forests, Inc., P.O. Box 340, Indiana, PA 15701
Phone: 724-465-5685; Fax: 724-465-9893

PA006 Pikes Peak Nurseries, R.D. 1, Box 75, Penn Run, PA 15765
Phone: 412-463-7747, 800-787-6730; Fax: 412-463-0775

PA007 Pine Grove Nursery, R.D. 3, Box 146, Clearfield, PA 16830
Phone: 814-765-2363, 800-647-1727

PA008 Ernst Conservation Seeds, 9006 Mercer Pike, Meadville, PA 16335
Phone: 814-425-7276, 800-873-3321; Fax: 814-425-2228

Pennsylvania (cont.)

PA009 Octoraro Native Plant Nursery, 6126 Street Rd.,
Kirkwood, PA 17536
Phone: 717-529-3160; Fax: 717-529-4099

PA010 Wetland Supply Co., 1633 Gilmar Rd., Apollo, PA 15613
Phone: 724-327-1830; Fax: 724-733-3527

PA011 Carino Nurseries, P.O. Box 538, Indiana, PA 15701
Phone: 724-463-3350, 800-223-7075; Fax: 724-463-3050

PA012 Sylva Native Nursery & Seed Co., 1683 Sieling Farm Road,
New Freedom, PA 17349
Phone: 717-227-0486; Fax: 717-227-0484

PA013 Hanchar's Superior Trees, RD 1, Box 118, Mahaffey, PA 15757
Phone: 814-277-6674

PA014 Beachly Hardy Agri Biotech, 454 Railroad Ave., P.O. Box 3147,
Shiremanstown, PA 17011
Phone: 717-737-4529

PA015 Seed, Inc., 307 Horsham Rd., Horsham, PA 19044
Phone: 215-675-2186

South Carolina

SC001 Carolina Nurseries, 739 Gaillard Rd., Moncks Corner, SC 29461
Phone: 843-761-8181, 800-845-2065

SC002 Wayside Gardens, 1 Garden Lane, Hodges, SC 29695
Phone: 800-845-1124; Fax: 800-817-1124

SC003 Woodlanders, Inc., 1128 Colleton Ave., Aiken, SC 29801
Phone: 803-648-7522; Fax: 803-648-7522

SC004 Park Seed Wholesale, 1 Parkton Ave., Greenwood, SC 29647-0002
Phone: 800-845-3366; Fax: 800-209-0360

SC005 AQUA-Tech Farms, 462 Juniper St., Neeses, SC 29107
Phone: 803-247-5697

SC006 National Wild Turkey Federation, P.O. Box 530,
Edgefield, SC 29824
Phone: 800-843-6983

South Dakota

SD001 Hansmeier & Son, Inc., Box 136, Bristol, SD 57219
Phone: 605-492-3611; Fax: 605-492-3254

SD002 The Sexauer Co., 100 Main Ave., P.O. Box 58, Brookings, SD 57006
Phone: 605-696-3600, 800-843-7929; Fax: 605-696-3610

SD003 Gurney's Seed & Nursery Co., 110 Capital St., Yankton, SD 57079
Phone: 605-665-1930, 605-665-1671; Fax: 605-665-9718

SD004 Milborn Seeds, Inc., 3127 Hwy 14 Bypass, Brookings, SD 57006
Phone: 605-697-6306

SD005 Den Besten Seed Co., Box 896, Platte, SD 57369
Phone: 605-337-3318

SD006 Pearl View Seeds, 21697 408 Ave., Cavour, SD 57324
Phone: 605-352-5933

SD007 Wilber's Seed, Inc., 800 N. Broadway, Box 41, Miller, SD 57362
Phone: 605-853-2414

Tennessee

TN001 Mill Creek Nursery Co., 6416 Short Mountain Rd.,
Smithville, TN 37166
Phone: 888-240-6567; Fax: 888-571-2163

TN002 Forest Nursery Co., Inc., 2362 Beersheba Hwy,
McMinnville, TN 37110
Phone: 931-473-2133, 931-473-4740; Fax: 931-473-2133

TN003 Warren County Nursery, Inc., 6492 Beersheba Hwy,
McMinnville, TN 37110
Phone: 931-668-8941; Fax: 931-668-2245

TN004 Triangle Nursery, Inc., 8526 Beersheba Hwy, McMinnville, TN 37110
Phone: 615-668-8022, 800-808-4769; Fax: 615-668-3297

TN005 Boyd Nursery Co., P.O. Box 71, McMinnville, TN 37111
Phone: 931-668-4747, 931-668-9898; Fax: 931-668-7646

TN006 Vernon Barnes & Son Nursery, 185 Kessey Ford Rd., P.O. Box 250,
McMinnville, TN 37110
Phone: 931-668-8576; Fax: 931-668-2165

TN007 Big Springs Nursery and Wildflowers, P.O. Box 513,
Beersheba Springs, TN 37305
Phone: 615-692-3604; Fax: 931-692-4023

TN008 Greenwood Nursery, P.O. Box 686, McMinnville, TN 37111
Phone: 800-426-0958; Fax: 931-668-2223

TN009 Lawson Wholesale Nursery, Inc., 6144 Beersheba Hwy,
McMinnville, TN 37110
Phone: 931-668-9514; Fax: 931-668-2094

TN010 Mountain Ornamental Nursery, P.O. Box 268, Altamont, TN 37301
Phone: 931-692-3424; Fax: 931-692-3331

TN011 Shahan Brothers Nursery, P.O. Box 876, Tullahoma, TN 37388
Phone: 615-455-3297; Fax: 615-393-2110

TN012 Flower City Nurseries, P.O. Box 75, Smartt, TN 37378
Phone: 931-668-4351, 931-668-3465; Fax: 931-668-3263

TN013 Sunlight Gardens, 174 Golden Lane, Andersonville, TN 37705
Phone: 800-272-7396, 423-494-8237; Fax: 423-494-7086

TN014 Schaefer Nursery, P.O. Box 62, 838 S. College St.,
Winchester, TN 37398
Phone: 931-967-4415, 931-967-8937; Fax: 931-967-6549

TN015 Joyce's Ground Covers, P.O. Box 102, McMinnville, TN 37111
Phone: 931-473-3263, 800-435-1142; Fax: 931-473-4628

TN016 Pleasant Cove Nursery, 2400 Old Rock Island Rd.,
Rock Island, TN 38581
Phone: 931-686-2215; Fax: 931-686-2362

TN017 Hillis Nursery Co., Inc., 92 Gardner Rd., McMinnville, TN 37110
Phone: 931-668-4364, 931-668-9125; Fax: 931-668-7432

TN018 Wanamaker Nursery, Inc., 12695 Beersheba Hwy,
McMinnville, TN 37110
Phone: 615-692-3763; Fax: 615-692-2397

Tennessee (cont.)

TN019 B&B Nursery, 6201 Beersheba Hwy, McMinnville, TN 37110
Phone: 931-668-8623

TN020 Ware's Nursery, P.O. Box 634, McMinnville, TN 37111
Phone: 615-668-9360

TN021 Circle M Farm, Rte. 6, Box 180, McMinnville, TN 37110
Phone: 615-668-8707

TN022 Tennessee Bush Farm, Inc., 11343 Beersheba Hwy, McMinnville, TN 37110-9638
Phone: 931-692-3624; Fax: 931-692-2397

TN023 Scott Bros. Nursery Co., P.O. Box 581, McMinnville, TN 37111
Phone: 931-473-2954, 800-435-1142; Fax: 931-473-4628

TN024 Dykes & Son Nursery & Greenhouse, 825 Maude Etter Rd., McMinnville, TN 37110
Phone: 931-668-8833, 931-668-7358; Fax: 931-668-2771

TN025 Native Gardens, 5737 Fisher Lane, Greenback, TN 37742
Phone: 423-856-0220; Fax: 423-856-0220

TN026 Trees By Touliatos, 2020 Brooks Rd., Memphis, TN 38116
Phone: 901-346-8065

Texas

TX001 Frontier Seed Co., 413 S. Avenue D, P.O. Box 177, Abernathy, TX 79311
Phone: 800-872-0522

TX002 Turner Seed, 211 CR 151, Breckenridge, TX 76424
Phone: 800-722-8616, 254-559-2065; Fax: 254-559-5024

TX003 Texas Forest Service, West Texas Nursery, Rt. 3, Box 216, Lubbock, TX 79401
Phone: 806-746-5801

TX004 Madrone Nursery, 2318 Hilliard Rd., San Marcos, TX 78666
Phone: 512-353-3944

TX005 Bamert Seed Co., Rt. 3, Box 1120, Muleshoe, TX 79347
Phone: 806-272-5506, 800-262-9892; Fax: 806-272-5509

TX006 Joe Moore, Rt. 1, Box 247, Gustine, TX 76455
Phone: 915-667-7356

TX007 Wildseed Farms, 1101 Campo Rosa Rd., P.O. Box 308, Eagle Lake, TX 77434
Phone: 800-848-0078; Fax: 409-234-7407

TX008 Bob Wells Nursery, P.O. Box 606, Lindale, TX 75771
Phone: 903-882-3550

TX009 Douglas W. King Company, P.O. Box 200320, San Antonio, TX 78220
Phone: 512-661-4191; Fax: 512-661-8972

TX010 Warner Bros. Seed Co., P.O. Box 1877, Hereford, TX 79045
Phone: 806-364-4470

TX011 Native American Seed, 127 N. 16th St., Junction, TX 76849
Phone: 915-446-3600; Fax: 915-446-4537

Texas (cont.)

TX012 Garrison & Townsend, Inc, P.O. Drawer 2420, Hereford, TX 79045
Phone: 806-364-0560; Fax: 806-364-3103

TX013 Texas Pecan Nursery, P.O. Box 306, Chandler, TX 75758
Phone: 903-849-6203

TX014 Texas Forest Service, Indian Mound Nursery, P.O. Box 617, Alto, TX 75925-0617
Phone: 409-858-4202

TX015 Dallas Nurseries, 2552 S. Stemmons, Lewisville, TX 75067
Phone: 214-316-2803, 800-235-6252

TX016 Robinson Seed Farms, 1113 Jefferson, Plainview, TX 79072
Phone: 806-293-4959

TX017 W.H. Anton Seed Co., Inc., P.O. Box 667, Lockhart, TX 78644
Phone: 512-398-2433

Utah

UT001 Granite Seed, 1697 W. 2100 North, Lehi, UT 84043
Phone: 801-768-4422, 801-531-1456; Fax: 801-768-3967

UT002 Progressive Plants, 9180 S. Wasatch Blvd., Sandy, UT 84093
Phone: 801-942-7333, 888-942-7333; Fax: 801-942-7383

UT003 Lone Peak Conservation Nursery, 271 W. Bitterbrush Lane, Draper, UT 84020
Phone: 801-571-0900

UT004 Stevenson Intermountain Seed, Box 2, Ephraim, UT 84627
Phone: 435-283-6639

UT005 Global Seed Co., P.O. Box 203, Gunnison, UT 84634
Phone: 435-528-3234

UT006 Maple Leaf Industries, Inc., 480 S. 50 East, Ephraim, UT 84627
Phone: 435-283-4701

Virginia

VA001 Naturescapes, 1581 Hosier Rd., Suffolk, VA 23434
Phone: 757-539-4833; Fax: 757-539-4833

VA002 Ingleside Plantation Nurseries, P.O. Box 1038, Oak Grove, VA 22443
Phone: 804-224-7111, 410-778-5787; Fax: 410-778-0135

VA003 Riverbend Nursery, Inc., 1295 Mt. Elbert Rd. N.W., Riner, VA 24149
Phone: 540-763-3362, 800-638-3362; Fax: 540-763-2022

VA005 Bobtown Nursery, 16212 Country Club Rd., Melfa, VA 23410
Phone: 757-787-8484, 800-201-4714; Fax: 757-787-8611

Washington

WA001 Fancy Fronds, P.O. Box 1090, Gold Bar, WA 98251
Phone: 360-793-1472

WA002 Tissues & Liners, Inc., 13245 Woodinville-Redmond Rd., Redmond, WA 98052
Phone: 425-885-5050; Fax: 425-861-5412

WA003 Plants of the Wild, P.O. Box 866, Tekoa, WA 99033
Phone: 509-284-2848; Fax: 509-284-6464

Washington (cont.)

WA004 Modern Forage Systems, Inc., 3770 Aldergrove Rd.,
Ferndale, WA 98248
Phone: 360-366-4345, 800-972-1812

WA005 Davenport Seed Co., P.O. Box 187, Davenport, WA 99122
Phone: 509-725-1235, 800-828-8873; Fax: 509-725-7015

West Virginia

WV001 Wrenwood of Berkeley Springs, Rt. 4, Box 361,
Berkeley Springs, WV 25411
Phone: 304-258-3071

Wisconsin

WI001 Wildlife Nurseries, Inc., P.O. Box 2724, Oshkosh, WI 54903
Phone: 920-231-3780; Fax: 920-231-3554

WI002 J.W. Jung Seed Co., 335 S. High St., Randolph, WI 53957
Phone: 800-247-5864; Fax: 800-692-5864

WI003 McKay Nursery Co., P.O. Box 185, Waterloo, WI 53594
Phone: 414-478-2121; Fax: 414-478-3615

WI004 Evergreen Nursery Co., Inc., 5027 County TT, Door County,
Sturgeon Bay, WI 54235
Phone: 800-448-5691; Fax: 920-743-9184

WI005 Hauser's Superior View Farm, Rt. 1, Box 199, Bayfield, WI 54814
Phone: 715-779-5404

WI006 Little Valley Farm, 5693 Snead Creek Rd., Spring Green, WI 53588
Phone: 608-935-3324

WI007 Kester's Wild Game Food Nurseries, Inc., P.O. Box 516,
Omro, WI 54963
Phone: 920-685-2929, 800-558-8815; Fax: 920-685-6727

WI008 Prairie Ridge Nursery, 9738 Overland Rd., Mt. Horeb, WI 53572
Phone: 608-437-5245; Fax: 608-437-8982

WI009 Prairie Nursery, P.O. Box 306, Westfield, WI 53964
Phone: 608-296-3679; Fax: 608-296-2741

WI010 Milaeger's Gardens, 4838 Douglas Ave., Racine, WI 53402
Phone: 800-669-9956; Fax: 414-639-1855

WI011 McClure & Zimmerman, 108 W. Winnebago St., P.O. Box 368,
Friesland, WI 53935-0368
Phone: 800-883-6998, 920-326-4220; Fax: 800-692-5864

WI012 J & J Transplant Aquatic Nursery, P.O. Box 227,
Wild Rose, WI 54984-0227
Phone: 715-256-0059, 800-622-5055; Fax: 715-256-0039

WI013 Roots & Rhizomes, P.O. Box A, Randolph, WI 53956-0118
Phone: 800-374-5035; Fax: 800-374-6120

Wyoming

WY001 Wind River Seed, 3075 Lane 51 1/2, Manderson, WY 82432
Phone: 307-568-3361; Fax: 307-568-3364

WY002 Etheridge Seed Farms, 1950 Lane 11, Powell, WY 82435
Phone: 307-754-2371

Directory of Wetland Plant Vendors

Part 2: Species and Sources

Abies balsamea **Fir, balsam**

CA003p, CA008s, CT005p, IA001p, ID003p, MA001s, MI003b, MI004p, MN003p, MN006p, MN017p, MT007p, NY004s, OR006p, OR008p, OR011p, OR014p, PA005p, PA006p, PA007p, PA011p

Acer negundo **Boxelder**

AR002p, CA002p, CA004s, CA007p, CA009p, CA013p, CO003p, CO005p, DE002p, FL016p, ID003p, ID007p, IN001p, LA008s, MA001s, MA003p, MD002p, MN002p, MT001p, MT005p, MT006p, MT007p, NJ001p, NJ005p, NJ008p, NY004s, NY005p, OR006p, OR011p, OR014p, PA006p, PA009p, PA012p, TN003p, TN007p, TN024p, UT002p, VA004p, VA005p

Acer rubrum **Maple, red**

AL001p, AL002p, AR002p, CA004s, CO003p, CO005p, CT001p, DE001p, DE002p, FL001p, FL003p, FL004p, FL005p, FL009p, FL010p, FL012p, FL013p, FL014p, FL015p, FL016p, GA002p, IA001p, IA004p, ID007p, IL003p, IL005p, IL006p, IN001p, IN002p, KY001p, LA001p, LA008s, MA001s, MA003p, MD002p, MI002p, MI003b, MI004p, MN002p, MN003p, MN005p, MN006p, MN009p, MN012p, MN014p, MN020p, MO002p, MS002p, MT007p, NC001p, NC002p, NJ001p, NJ004p, NJ008p, NY003p, NY004s, NY006p, OH003p, OH004p, OH008p, OK001p, OK004p, OK005p, OR001p, OR003p, OR005p, OR007p, OR008p, OR009p, OR010p, OR014p, OR015p, OR016p, PA001p, PA003p, PA004p, PA005p, PA006p, PA007p, PA008s, PA009p, PA010p, PA012p, SC001p, SC003p, TN001p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN010p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, UT002p, VA002p, VA004p, VA005p, WI003p, WI012p

Acer rubrum* var. *drummondii **Maple, Drummond red**

CA004p, LA001p, LA002p, LA007p, LA008s, MS002p, NY004s, OK001p

Acer rubrum* var. *trilobum **Maple, trident red**

NJ005p

Acer saccharinum **Maple, silver**

AL002p, AR002p, CA004s, CO001p, CO005p, DE002p, FL015p, FL016p, IA001p, IA004p, ID003p, ID007p, IN001p, LA001p, LA008s, MA001s, MA003p, MD002p, MI002p, MI003b, MN002p, MN003p, MN005p, MN012p, MN014p, MN017p, MO002p, MT002p, MT005p, MT007p, NJ005p, NJ008p, NY004s, OH008p, OK001p, OK004p, OK005p, OK007p, OR003p, OR005p, OR015p, PA005p, PA009p, PA010p, PA012p, TN001p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN010p, TN012p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, TX008p, UT002p, VA004p, VA005p, WI002s, WI003p, WI012p

Acoelorraphe wrightii **Palm, Paurotis**

CA004s, FL003p, FL004p

Aconitum columbianum **Monkshood, Columbia**

IA002s

Acorus americanus **Sweetflag**

WI012b

Acorus calamus **Sweetflag**

CO004p, CO005p, CT002p, CT009p, IA003s, IL002s, IL004p, IN003p, KY002p, MA002p, MA003p, MA004p, MD001p, MD002p, MI001p, MN004b, MN009b, MN010p, MN013s, MT001p, NC003p, NC008p, NE003s, NE011p, NJ008p, OH006p, OH008p, OR014p, PA004p, PA008s, PA009p, PA010p, PA012p, SC001p, VA001p, VA004p, VA005p, WI001p, WI007p, WI008s, WI010p

Acrostichum danaeifolium **Fern, inland leather**

FL003p, FL004p, FL008p

Adiantum capillus-veneris **Fern, southern maiden-hair**

CA002p, CA006p, FL012p, FL013p, FL016p, NC005p, NY005p, OH005p, OR014p, SC003p, WA001p

Aegopodium podagraria **Goutweed, Bishops**

AR002p, CO001p, CO003p, CO005p, CT002p, CT003p, GA001p, IL003p, IL004p, IL006p, IN001p, MI001p, MI004p, MN002p, MN003p, MN006p, MN010p, MN014p, MN020p, NJ001p, NE011p, NY002p, NY005p, OH003p, OH008p, OR008p, OR011p, OR014p, SC002p, TN015p, TN023p, UT002p, VA003p, WA002p, WI005p

Agalinis purpurea **False-foxglove, large purple**

MN009s

Agalinis tenuifolia **False-foxglove, slender**

KY002p, MN009s

Agarista populifolia **Hobblebush, Florida**

FL012p, FL015p, SC003p

Agrimonia parviflora **Groovebur, small-flower**

PA008s

Agrostis capillaris **Bentgrass, colonial**

MN018s, PA008s, UT001s

Agrostis gigantea **Redtop**

CO007s, CO009s, ID004s, ID005s, IL002s, KS001s, MA002p, MO001s, MO006s, MO007s, MT003s, MT004s, NE005s, NJ003s, NJ008p, PA004p, PA008s, PA010b, SD002s, UT001s, UT004s, VA004p, WI001s, WY001s

Agrostis hyemalis **Bentgrass, winter**

MT004s

Agrostis stolonifera **Bentgrass, spreading**

CA017b, NE002s, NJ003s, PA008s, PA010b, SD002s, UT001s

Aletris lutea **Colicroot, yellow**

FL015p

Alisma lanceolatum **Water plantain, American**

NC005p

Alisma plantago-aquatica **Water plantain, broad-leaf**
CA017b, CO005p, CT009b, IA003s, MA002p, MA003p, MN013s, NC005p, NC008p, PA004p, PA008s, PA010b, PA012p, WI001p, WI007p, WI008s

Alisma subcordatum **Water plantain, subcordate**
IL004s, KY002p, MN004s, MN009b, WI012b

Allenrolfea occidentalis **Iodinebush**
CA007p

Allium geyeri **Onion, Geyer's**
OR014p

Allium schoenoprasum **Chives**
CA004s, CT002p, CT008s, FL012p, IA004s, ID004s, IL004p, KY003p, ME001s, ME003s, MI001p, MN002p, MN010p, MN014s, NC005p, NE003s, NE011p, NJ007s, NY004s, OH005b, OH006p, PA002s, SC004b, SD003b, TX007s, VA003p, WI002s, WI005p, WV001p

Allium validum **Onion, Pacific**
CA007p

Alnus glutinosa **Alder, European**
CO003p, CO005p, IN001p, MA001s, MN002p, MT002p, MT007p, NE003s, NY004s, OH003p, OH008p, OR011p, OR014p, PA004p, PA005p, PA006p, TN022p, VA005p, WI003p

Alnus incana **Alder, speckled**
CA004s, CO004p, CO005p, ID002s, MA001s, MA003p, MT001p, NY004s, OR014p, UT002p, UT004s, VA005p

Alnus incana* ssp. *rugosa **Alder, speckled**
NJ005p, NJ008p, NY004s, OR016p, PA004p, PA005p, PA008s, PA009p, PA010p, PA012p, VA004p

Alnus incana* ssp. *tenuifolia **Alder, thinleaf**
CA007p, CA008s, CO001p, CO003p, CO004p, CO005p, ID001p, ID003p, MA001s, MT001p, MT002p, MT005p, MT006p, NY004s, OR014p, PA010p, UT003p, WA003p

Alnus maritima **Alder, seaside**
OR014p

Alnus rhombifolia **Alder, white**
CA002p, CA004s, CA005p, CA007p, CA008s, CA012p, CA017b, ID002s, MA001s, MT001p, NY004s, OR014p

Alnus rubra **Alder, red**
CA002p, CA004s, CA008s, CA017b, ID002s, MA001s, NY004s, OR008p, OR009p, OR014p, VA005p, WA003p

Alnus serrulata **Alder, hazel**
AR004p, FL009p, ID002s, KY002p, MA001s, MD002p, NJ001p, NJ005p, NJ008p, NY004s, PA009p, PA010p, PA012p, VA001p, VA004p, VA005p

Alnus viridis* ssp. *crispa **Alder, mountain**
MA001s

Alnus viridis* ssp. *sinuata **Alder, Sitka**
ID001p, ID002s, ID003p, MA001s, MT001p, NY004s, OR014p, WA003p

Alocasia macrorrhizos **Taro, giant**
FL016p, LA003p

Alopecurus arundinaceus **Foxtail, creeping**
CO007s, CO009s, ID006s, KS001s, MN019s, MT003p, MT004s, MT008s, MT009s, MT010s, MT011s, MT012s, MT013s, MT014s, NE005s, NE006s, NE010s, PA008s, PA010b, SD001s, SD002s, SD004s, SD005s, SD006s, SD007s, UT001s, UT004s, UT005s, UT006s, WY001s, WY002s

Alopecurus geniculatus **Foxtail, water**
CA017b

Alopecurus pratensis **Foxtail, meadow**
CA017b, CT002p, IL002s, MT004s, NE011p, OR014p, PA008s, PA010b, UT001s, WA002p, WI010p, WY001s

Althaea officinalis **Marshmallow, common**
NY004s, OH006p

Ambrosia trifida **Ragweed, great**
IL002s

Amorpha fruticosa **Indigobush, false**
AR005p, CA004s, CA005p, CO001p, CO004p, CO005p, FL015p, GA002p, IL004b, KY002p, LA002p, MA001s, MN009b, NE011p, NJ005p, NY004s, OR014p, PA008s, PA010p, UT003p

Ampelaster carolinianus **Aster, climbing**
CT002p, FL008p, FL014p, FL015p, FL016p, NC004p, NC005p, NJ001p, NY002p, NY005p, OR014p, SC003p, TN025p

Ampelopsis arborea **Peppervine**
AR005p, LA008s, OR014p

Amphicarpaea bracteata **Hogpeanut, American**
MA004p

Amsonia tabernaemontana **Bluestar, eastern**
AR005p, IL004p, KY002p, MA004p, MN002p, MN010p, NC004p, NC005p, NE003s, NE011p, NJ001p, NJ007s, OH007p, OH008p, OR014p, SC001p, SC003p, SD003p, TN013p, TN025p, VA003p, WI010p, WI013p

Anagallis arvensis **Pimpernel, scarlet**
CO006s, ID004s

Andromeda polifolia **Rosemary, bog**
CT001p, CT004p, ME002p, MN002p, MN006p, OH003p, OR008p, OR011p, OR014p, SC002p

Andropogon glomeratus **Bluestem, bushy**
MD002p, NC008p, NJ005p, SC001p, TN025b, TX004p

Anemone canadensis **Thimbleweed, Canada**
CO005p, IL004p, MA004p, MN001p, MN004b, MN009b, MN011s, NJ005p, OR004p, OR014p, TN013p, WI006p, WI008b, WI009p

Anemopsis californica **Yerba mansa**
CA002p, CA007p, CA009p, CA010s

Angelica atropurpurea **Angelica, purplestem**
IL004b, MN007s, MN009b, NE003s, NY004s, PA008s, WI008s, WI009s

Annona glabra **Apple, pond**
FL003p, FL004p, FL008p

Apios americana **Groundnut**
MA004p, OR014p, PA008s

Apium graveolens **Celery**
CT008s, OH005p, ME001s, ME003s, PA002p, WI002s

Aplectrum hyemale **Adam and Eve**
KY003p, NJ001p

Aquilegia canadensis **Columbine, red**
AR002p, AR005p, CA011s, CO002s, CO003p, CO005p, CO006s, CT002p, CT004p, FL012p, FL014p, FL015p, ID002s, IL001s, IL004b, IN003p, KY002b, MA004p, MN001p, MN002p, MN004b, MN006p, MN009b, MN010p, MO003b, MT015p, NC004p, NC005p, NC006b, NE003s, NE011p, NJ001p, NJ005p, NJ007s, NY005p, OH005p, OR004p, OR014p, PA008s, SC001p, SC003p, TN003p, TN006p, TN010p, TN013p, TN015p, TN017p, TN019p, TN022p, TN023p, TN024p, TN025p, TX004p, UT001s, VA003p, WI006p, WI008b, WI009p, WI010p

Aquilegia chrysantha **Columbine, golden**
AZ004s, CA006p, CO001p, CO002s, CO003p, CT002p, MN002p, NC005p, NE003s, NE011p, NJ007s, NY005p, OH005p, OH007p, OR014p, SC001p, TN013p, UT001s, VA003p, WI010p

Aquilegia chrysantha* var. *hinckleyana **Columbine, Hinckley's golden**
OR014p, TX004p

Aquilegia coerulea **Columbine, Colorado blue**
CO005p, CO006p, CT002p, FL012p, ID002s, ID004s, MT001p, NE003s, NE011p, NJ007s, NY002p, NY004s, OH005b, OR014p, UT001s, UT002p, WA003p, WY001s

Aquilegia eximia Columbine **Columbine, Van Houtte's**
CA002p

Aquilegia formosa* var. *formosa **Columbine, western**
CA002p

Aralia californica **Spikenard, California**
CA017b

Aralia spinosa **Walkingstick, devils**
FL014p, LA008s, MA001s, MD002p, NY004s, OH008p, PA008s, SC003p

Argentina anserina **Silverweed**
CA017p, MN001p, OR009p

Arisaema dracontium **Dragon, green**
FL015p, NC005p, NY005p, TN013p, TN024p, TN025p

Arisaema triphyllum **Jack in the pulpit, swamp**
FL015p, IA004p, IL001s, IL004p, KY002p, KY003p, MN004b, MN006p, MN009p, NC004p, NC005p, NC006p, NC007p, NE001p, NE011p, NY004s, NY005p, OR004p, PA008s, PA010p, SC002p, TN002p, TN003p, TN006p, TN010p, TN013p, TN015p, TN017p, TN019p, TN022p, TN023p, TN024p, TN025p, WI002p, WI006p, WI008p, WI009b, WI012p

Arnica chamissonis **Arnica, leafy**
CO005p

Artemisia cana **Sagebrush, silver**
CA002p, CO001p, CO004p, CO005p, ID002s, MT001p, MT004s, NE003s, UT001s, WY001s

Artemisia douglasiana **Sagewort, Douglas'**
CA002p, CA005p, CA007p, CA009p, CA010s, CA012p

Aruncus dioicus **Bride's feathers**
AR005p, CO001p, CT002p, CT004p, GA001p, IL004p, KY002p, KY003p, MI001p, MN002p, MN006p, MN010p, MN020p, NC004p, NC005p, NE003s, NE011p, NJ001p, NJ007s, NY002p, NY004s, NY005p, OH002p, OH005s, OH007p, OH008p, OR004p, OR014p, SC001p, SC004s, TN010p, VA003p, WI002p, WI010p, WI013p

Arundinaria gigantea **Cane, giant**
CA001p, KY002p, MA004p, OH008p

Arundo donax **Reed, giant**
MA004p, NC008p, NE011p, OH003p, OH008p, OR011p, VA005p

Asarum lemmonii **Wildginger, Lemmon's**
CA007p

Asclepias incarnata **Milkweed, swamp**

AR002p, AR005p, CO003p, CO004p, CO005p, CT002p, CT009s, GA001p, IA003s, IL001s, IL004b, IN003p, KY002b, MA003p, MA004p, MD002p, ME001s, MI001p, MN001p, MN004b, MN007s, MN009b, MN010p, MN011s, NC004p, NC005p, NE002s, NE003s, NE011p, NJ001p, NJ005p, NJ008p, NY004s, NY005p, OH005p, OH007p, OH008p, OH010p, OR011p, OR014p, PA004p, PA008s, PA009p, PA010p, PA012p, SC001p, SC003p, SC004b, TN013p, TN025p, VA003p, VA004p, VA005p, WI001p, WI005p, WI006p, WI007p, WI008b, WI009b, WI010p, WI012b, WI013p

Asclepias speciosa **Milkweed, showy**

AR005p, CA007p, ME001s, MN009s, OR014p

Aster ascendens **Aster, California**

CA002p, CA012p

Aster lanceolatus* ssp. *lanceolatus var. lanceolatus **Aster, panicled**

AR005p, MN009p, NC005p, NJ005p, WI008s, WI012b

Aster lateriflorus **Aster, calico**

CT002p, CT004p, IL004b, IN003p, KY002p, MA004p, MI001p, MN001p, MN004b, MN009s, MN010p, NC004p, NC005p, NE011p, NJ001p, NY004s, OH002p, OH007p, OR011p, OR014p, PA008s, SC001p

Aster lateriflorus* var. *lateriflorus **Aster, small white**

CO005p, MN010p, NC005p, NJ005p, OR014p

Aster novae-angliae **Aster, New England**

AR005p, CA010s, CA011s, CO002s, CO003p, CO005p, CO006s, CT003p, CT004p, CT009p, FL014p, GA001p, IA003s, ID002s, IL001s, IL004b, IN003p, KY002b, MA003p, MA004P, ME001s, ME003p, MI001p, MN002p, MN004b, MN006p, MN007s, MN009b, MN010p, MN011s, MO003b, NC005p, NE002s, NE003s, NE011p, NJ001p, NJ002p, NJ008p, NY004p, OH002p, OH005b, OH008p, OK007p, OK002s, OR014p, PA004p, PA008s, PA009p, PA010b, PA012p, SC001p, SC002p, SC003p, TN013p, TN025b, TX007s, UT001s, UT002p, VA002p, VA003p, VA004p, WI005p, WI006p, WI008b, WI009b, WI010p, WI012b, WI013p, WY001s

Aster novi-belgii **Aster, New York**

CA006p, CO001p, CO002s, CO003p, CO005p, CO006s, FL012p, GA001p, MD002p, MI001p, MN006p, MN010p, NC005p, NE011p, NJ001p, NJ002p, NJ005p, NJ008p, OH002p, OH005p, OH007p, OH008p, OH010p, OR014p, PA004p, PA008s, PA009p, PA010b, PA012p, VA002p, VA004p, WV001p

Aster paludosus **Aster, southern swamp**

AR005p

Aster praealtus **Aster, willowleaf**

AR005p, IL004p, MN009s, NC005p

Aster puniceus **Aster, swamp**

IL004p, IN003p, MA003p, MN004b, MN009b, NC005p, NJ001p, PA008s, PA009p, PA010b, PA012p

Aster puniceus* var. *firmus **Aster, shining**
AR005p

Astragalus canadensis **Milkvetch, Canada**
IL004b, KY002p, MN004b, MN007s, MN009b, MN011s, MN013s, WI008s, WI009s

Atriplex hortensis **Orache, garden**
NE003s, NJ007s

Atriplex lentiformis **Saltbush, lensfruit**
AZ002p, CA002p, CA004s, CA005p, CA006p, CA007p, CA010s, CA011s, CA013p, UT001s

Atriplex patula **Saltbush, halberdleaf**
CA017b, IL002s

Atriplex semibaccata **Saltbush, Australian**
CA004s, CA009p, CA010s, CA011s, UT001s

Atriplex tridentata **Saltbush, basin**
UT001s, UT004s, WY001s

Avicennia germinans **Mangrove, black**
FL002p, FL005p, FL006p, FL016p, LA006p

Axonopus fissifolius **Carpetgrass, common**
GA003s

Azolla caroliniana **Mosquitofern, Carolina**
CT004p, MA004p, NJ001p

Azolla filiculoides **Mosquitofern, Pacific**
CA017p

Baccharis angustifolia **False willow, saltwater**
FL016p, SC003p

Baccharis douglasii **False willow, Douglas'**
CA002p, CA007p

Baccharis emoryi **False willow, Emory's**
CA010s

Baccharis halimifolia **False willow, eastern**
CT002p, FL016p, MD002p, ME002p, NC004p, NJ001p, NJ005p, NJ008p, NJ009p, NY004s, NY005p, OR014p, PA009p, PA012p, SC001p, SC003p, VA001p, VA004p, VA005p

Baccharis salicifolia **Mule's fat**
CA002p, CA007p, CA010s

Bacopa caroliniana **Waterhyssop, Carolina**
FL016p, FL017p, NC003p

Bacopa monnieri **Waterhyssop, herb of grace**
FL016p, FL017p

Barbarea vulgaris **Yellowrocket, garden**
IL002s

Beckmannia syzigachne **Sloughgrass, American**
CA017b, CO004p, CO005p, PA010p, UT001s

Berchemia scandens **Supplejack, Alabama**
OR014p

Betula nana **Birch, dwarf**
CA003p, CO003p, CO004p, CO005p, MA001s, MT001p, NY004s, PA010p

Betula nigra **Birch, river**
AL001p, AL002p, AL006p, AL007p, AR002p, AR004p, AR005p, CA004s, CT001p, CT004p,
CT006p, DE002p, FL001p, FL005p, FL009p, FL010p, FL012p, FL014p, FL015p, FL016p,
GA002p, IA001p, ID003p, ID007p, ID008p, IN001p, IN002p, KY001p, LA001p, LA003p,
LA005p, LA007p, LA008s, MA001s, MA003p, MD002p, MI002p, MI003b, MI004p, MN002p,
MN003p, MN005p, MN006p, MN009p, MN012p, MN020p, MO002p, MS001p, MS002p, MT005p,
MT007p, MT015p, NC001p, NC002p, NJ001p, NJ002p, NJ004p, NJ005p, NJ008p, NY003p,
NY004s, NY006p, OH003p, OH004p, OH008p, OK001p, OH004p, OH008p, OK001p, OK004p,
OK005p, OR001p, OR002p, OR003p, OR005p, OR009p, OR011p, OR014p, OR015p, OR016p,
PA003p, PA004p, PA005p, PA006p, PA007p, PA009p, PA010p, PA012p, SC002p, TN001p,
TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN010p, TN011p,
TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, UT002p,
VA002p, VA004p, VA005p, WI002s, WI003p, WI004p, WI012p

Betula occidentalis **Birch, spring**
CA002p, CA012p, CO004p, CO005p, ID002s, ID003p, ID007p, MN002p, MT001p, MT002p,
MT015p, NY004s, OR014p, PA010p, UT002p, WA003p

Betula pumila **Birch, bog**
ID003p, MN009p, NY004s, OR011p, OR014p

Bidens aristosa **Beggarticks, bearded**
AR004p, KY002b, NC006s, PA008s

Bidens cernua **Beggarticks, nodding**
MN009s, PA008s, PA012p

Bidens connata **Beggarticks, purplestem**
MN009s

Bidens coronata **Beggarticks, crowned**
MN009s

Bidens frondosa **Beggarticks, devil's**
IL002s, PA008s, PA010b, PA012p, WI008s

Bidens vulgata **Beggarticks, big devils**
WI008s

Bignonia capreolata **Crossvine**
AR002p, FL012p, KY002p, LA002p, LA003p, MA004p, NC004p, NC005p, NY005p, OK001p, OR014p, SC001p, SC002p, SC003p

Blechnum serrulatum **Fern, toothed midsorus**
FL005p, FL006p

Boltonia asteroides **White doll's daisy**
CO001p, CO003p, CO005p, CT002p, CT003p, CT004p, IL004p, KY002b, MA004p, MI001p, MN002p, MN009s, MN010p, NC004p, NC005p, NE011p, NY002p, NY005p, OH002p, OH005b, OH007p, OH008p, OR004p, OR011p, OR014p, SC001p, SC004s, TN013p, VA003p, WI010p, WI013p

Borrichia arborescens **Tree seaside tansy**
FL004p, FL005p, FL016p

Borrichia frutescens **Bushy seaside tansy**
FL002p, FL005p, FL006p, FL008p

Boykinia aconitifolia **Brookfoam, Alleghany**
NC005p

Boykinia major **Brookfoam, mountain**
OR004p, OR014p

Brasenia schreberi **Watershield**
NJ005p, WI012p

Bromus ciliatus **Brome, fringed**
MN004b, MN009s

Bromus latiglumis **Brome, earlyleaf**
IL004p

Brugmansia suaveolens **Angel's-tears**
SC003p

Brunnichia ovata **American buckwheat vine (Redvine)**
AR005p

Bucida buceras **Gregorywood**
FL005p, FL006p

Butomus umbellatus **Flowering-rush**
NC003p, NC008p, OH008p, PA010b, WI001p, WI007p, WI012p

Cabomba caroliniana **Fanwort, Carolina**
CT004p, NC003p, NC008p

Calamagrostis bolanderi **Reedgrass, Bolander's**
CA007p

Calamagrostis canadensis **Reedgrass, bluejoint**
CO004p, CO005p, IL004p, KY002b, MA002p, MA003p, MN004b, MN007s, MN009b, NJ008p, OR009p, PA008s, PA009p, PA010b, PA012p, UT001s, VA004p, VA005p, WI001p, WI008b, WI012b

Calamagrostis coarctata **Reedgrass, arctic**
NJ005p

Calamagrostis nutkaensis **Reedgrass, Pacific**
CA006p, CA007p, CA009p, CA013p

Calamagrostis stricta **Reedgrass, slimstem**
PA010p

Calamagrostis stricta ssp. inexpansa **Reedgrass, northern**
CO004p, CO005p

Calla palustris **Water arum (wild calla)**
MN001p, NC003p, PA004p, PA010p, WI001p, WI012p

Callicarpa dichotoma **Beautyberry, purple**
MA001s, NC004p, NC005p, NC007p, NE003s, NE011p, NY004s, NY005p, OH008p, OR005p, OR011p, OR014p, SC001p, SC003p, VA002p, VA005p

Calophyllum antillanum **Calophyllum, Antilles**
FL005p

Caltha palustris **Marsh marigold, yellow**
CT002p, CT004p, GA001p, IL001s, IL004p, KY002p, MA003p, MN001p, MN004b, MN006p, MN009b, NC003p, NE011p, NJ001p, NY005p, OH007p, OH008p, OR004p, PA004p, PA008s, PA010p, PA012p, SD003p, TN013p, VA005p, WI006p, WI008b, WI009p, WI010p, WI012p

Calydorea coelestina **Ixia, Bartram's**
SC003p

Camassia leichtlinii **Camas, large (Leichtlin's Camassia)**
GA001p, OR004p, OR014p, WI002p

Camassia quamash **Camas, small**
CA002p, CA017b, GA001p, ID002s, ID004s, ME003p, OR009p, OR014p, WA003p, WI002p

Camassia scilloides **Camas, Atlantic**
KY002p, MN009b, NC005p

Canna flaccida **Bandanna of the Everglades**
CT004p, FL005p, FL010p, FL015p, FL016p, FL017p, MS003p, SC003p

Canna glauca **Maraca amarilla**
NY004s

Canna indica **Shot, Indian**
MS003p

Canna x generalis **Canna lily**
AR002p, FL012p, IA004p, IL005p, MD001p, MI005p, NY004s, OH005b, OH010p, OK001p, OR011p, PA002p, SC001p, SC002p, SC003s, SD003p, WI002p, WI011p

Cardamine pratensis **Bittercress, meadow cuckoo flower**
CT002p, NC005p, NY005p

Carex abscondita **Sedge, thicket**
MA004p

Carex alata **Sedge, broadwing**
NJ005p

Carex albida **Sedge, whitetinge**
CA006p

Carex albolutescens **Sedge, greenwhite**
NJ005p

Carex alopecoidea **Sedge, foxtail**
MN009b

Carex amplifolia **Sedge, bigleaf**
CA007p

Carex annectens **Sedge, yellowfruit**
CT009s, IL004p, MN009b, NC005p, PA008s

Carex aperta **Sedge, Columbian**
OR008p

Carex aquatilis **Sedge, water**
CO003p, CO004p, CO005p, ID002s, MT001p, PA008s, PA010b, UT001s, UT003p, WI008s, WI012b

Carex atherodes **Sedge, wheat (slough sedge)**
IL004p

Carex athrostachya **Sedge, slenderbeak**
MT001p

Carex atlantica **Sedge, prickly bog**
NJ005p

Carex atlantica* ssp. *capillacea **Prickly bog sedge**
NJ005p

Carex barbarae **Sedge, Santa Barbara**
CA007p, CA009p, CA013p, CA017p

Carex bebbii **Sedge, Bebb's**
CO004p, CO005p, IL004b, MT001p, PA008s, PA010p, UT001s, WI008s, WI009s, WI012b

Carex bicknellii **Sedge, Bicknell's**
IL004p

Carex bolanderi **Sedge, Bolander's**
CA017b

Carex brittoniana **Sedge, Britton's**
TX004p

Carex canescens **Sedge, silvery**
NJ005p

Carex cephalophora **Sedge, oval-leaf**
IL004p

Carex comosa ***Sedge, bearded***
CT009b, IL004b, MA003p, MN004b, MN009b, NC005p, PA008s, PA010p, WI001b, WI007p, WI008s, WI012b

Carex conjuncta **Sedge, soft fox**
IL004b

Carex crinita ***Sedge, fringed***
CO011b, IL004b, IN003p, KY002p, MA002p, MA003p, MA004p, MD002p, MN004b, MN009s, NC005p, NJ005p, NJ008p, PA004p, PA008s, PA009p, PA010p, PA012p, VA001p, VA004p, WI008s

Carex cristatella **Sedge, crested**
IL004b, IN003p, NC005p, PA008s

Carex crus-corvi **Sedge, ravenfoot**
IL004b, NC005p, TX004p

Carex densa **Sedge, dense**
MT001p

Carex disperma **Sedge, softleaf**
CA002p

Carex emoryi **Sedge, Emory's**
PA008s, TX004p

Carex feta — **Sedge, greensheath**
MT001p

Carex flaccosperma — **Sedge, thinfruit**
NC005p

Carex flava — **Sedge, yellow**
WI008s

Carex frankii — **Sedge, Frank's**
IL004b, IN003p, KY002p, NC005p, NJ005p

Carex granularis — **Sedge, limestone meadow**
IN003p

Carex grayi — **Sedge, Gray's**
IL004b, IN003p, MA004p, MN009s, NC005p, NE011p, OR014p, PA008s, WI008s

Carex gynandra — **Sedge, nodding**
CT009s, PA008s

Carex haydeniana — **Sedge, cloud**
MT001p

Carex haydenii — **Sedge, Hayden's**
IL004p

Carex hystericina — **Sedge, bottlebrush**
CO005p, IL004p, MN009b, PA008s, PA010b, WI008s, WI009s, WI012b

Carex intumescens — **Sedge, greater bladder**
CT009b, MD002p, NJ005p, PA008s, WI008s

Carex lacustris — **Sedge, hairy (lake sedge)**
CT009p, IL004p, IN003p, MA003p, MN009s, PA008s, PA010b, WI001p, WI007p, WI008s, WI009s, WI012b

Carex lanuginosa — **Sedge, wooly**
CA012p, CO003p, CO004p, CO005p, MT001p, PA010p, WI008s

Carex lasiocarpa — **Sedge, woolly-fruit**
WI012p

Carex lonchocarpa — **Sedge, southern long**
NC005p

Carex longii — **Sedge, Long's**
NJ005p

Carex lupulina — **Sedge, hop**
CT009b, IL004b, MA003p, MN009s, NJ005p, PA008s, WI012p

Carex lurida **Sedge, shallow**
AR005p, CT009b, IN003p, MA002p, MA003p, MA004p, MD002p, NC005p, NJ005p, NJ008p, PA008s, PA009p, PA010b, PA012p, VA001p, VA004p, VA005p, WI008s

Carex lyngbyei **Sedge, Lyngbye's**
CA017b, OR008p, OR009p

Carex microptera **Sedge, smallwing**
CO004p, MT001p, PA010p, UT003p

Carex mitchelliana **Sedge, Mitchell's**
NJ005p

Carex muskingumensis **Sedge, Muskingum**
AR005p, CT002p, GA001p, IL004b, NC005p, NC008p, NE011p, NJ001p, OH008p

Carex nebrascensis **Sedge, Nebraska**
CA017b, CO004p, CO005p, MT001p, PA010p, UT001s

Carex nigra **Sedge, smooth black**
AR005p, NJ001p, OH008p, OR008p, OR011p, OR014p, WA002p

Carex normalis **Sedge, greater straw**
IL004p

Carex obnupta **Sedge, slough**
CA017b, OR008p, OR009p

Carex praegracilis **Sedge, clustered field**
CA002p, CA006p, CA007p, CA009p

Carex prairea **Sedge, prairie**
MN009s

Carex prasina **Sedge, drooping**
PA008s

Carex projecta **Sedge, necklace**
MN009s, NC005p

Carex pseudocyperus **Sedge, cypresslike**
WI008s

Carex retrorsa **Sedge, knotsheath**
IL004b, MN009b, PA010p, WI008s

Carex rostrata **Sedge, beaked**
CA017b, CO004p, CO005p, MT001p, NJ005p, OR008p, PA004p, PA010p, UT001s, UT003p, WI008s, WI012p

Carex sartwellii **Sedge, Sartwell's**
IL004b

Carex scoparia **Sedge, pointed broom**
CT009b, IL004p, MA002p, MN004b, MN009b, NJ005p, PA008s, WI008s, WI012b

Carex scopulorum **Sedge, mountain**
CO004p, PA010p

Carex senta **Carex, swamp**
CA002p

Carex seorsa **Sedge, weak stellate**
PA008s

Carex shortiana **Sedge, Short's**
IL004b, NC005p

Carex simulata **Sedge, analogue**
CO005p

Carex spectabilis **Sedge, showy**
MT001p

Carex squarrosa **Sedge, squarrose**
NJ005p

Carex sterilis **Sedge, dioecious**
MN009s

Carex stipata **Sedge, stalkgrain**
CO004p, IL004p, IN003p, MN009s, NC005p, NJ008p, OR009p, PA008s, PA010p, VA004p, WI008s, WI012b

Carex stricta **Sedge, upright**
CT002p, CT004p, CT009b, IL004p, IN003p, MA002p, MA003p, MA004p, MD002p, MN004b, MN009s, NC005p, NJ001p, NJ005p, NJ008p, PA004p, PA008s, PA009p, PA010p, PA012p, VA004p, VA005p, WI001p, WI007p, WI008s, WI012b

Carex tribuloides **Sedge, blunt broom**
CT009p, IL004p, NJ005p, PA008s

Carex trichocarpa **Sedge, hairyfruit**
WI008p

Carex tuckermanii **Sedge, Tuckerman's**
MN009s, WI008s

Carex typhina **Sedge, cattail**
MN009s, PA008s

Carex vesicaria **Sedge, western inflated**
MT001p, PA008s

Carex viridula **Sedge, little green**
WI008s

Carex vulpinoidea ***Sedge, fox***
CO005p, CT009b, IL004b, IN003p, KY002p, MA002p, MA003p, MN009b, NC005p, NJ005p, NJ008p, PA008s, PA009p, PA010b, PA012p, VA001p, VA004p, VA005p, WI008s, WI009s, WI012b

Carphephorus odoratissimus **Vanillaleaf**
FL015p

Carphephorus paniculatus **Chaffhead, hairy**
FL015p

Carya aquatica **Hickory, water**
FL001p, FL009p, FL010p, FL014p, FL015p, FL016p, LA002p, LA004p, LA008s, MS001p, NY004s, OR014p, SC003p

Carya illinoinensis **Pecan**
AL002p, AL007p, AR002p, GA002p, IA001p, IA004p, IL003p, IL005p, IL006p, LA005p, LA008s, MA001s, MN017p, MN020p, MO002p, MO004p, MO005p, MS001p, NY004s, OK004p, OK007p, OR014p, PA005p, SD003p, TN002p, TN003p, TN005p, TN006p, TN007p, TN008p, TN017p, TN018p, TN020p, TN021p, TN022p, TX004p, TX008p, TX013p, TX015p, VA005p

Carya laciniosa **Hickory, big shellbark**
FL001p, FL014p, GA002p, IA001p, IA004p, LA008s, MA001s, MO002p, MO004p, NY004s, PA012p, SC003p, SD003p, TN003p, TN007p, TN017p, TN018p, TN021p, TN022p

Carya myristiciformis **Hickory, nutmeg**
FL014p, LA008s, NY004s, OR014p, SC003p

Carya X lecontei **Pecan, bitter**
LA005p

Castilleja miniata **Indian paintbrush, giant red**
ID002s, ID004s

Castilleja minor ssp. minor **Indian paintbrush, lesser**
UT001s

Castilleja sulphurea **Indian paintbrush, sulphur**
UT001s

Celtis laevigata **Sugarberry**
FL001p, FL003p, FL005p, FL007p, FL009p, FL010p, FL014p, FL015p, FL016p, LA008s, MO002p, NC001p, NY004s, OR014p, TN003p

Celtis laevigata* var. *reticulata **Hackberry, netleaf**

ID002s, MT007p, NY004s, OR014p, UT003p

Cephalanthus occidentalis **Buttonbush, common**

AR004p, AR005p, CA002p, CA007p, CT004p, CT005p, CT009p, DE002p, FL001p, FL003p, FL010p, FL013p, FL014p, FL015p, FL016p, IL004b, IN003p, KY002p, LA002p, LA008s, MA001s, MA002p, MA003p, MA004p, MD002p, MN002p, MN009b, MO001s, MO002p, NC001p, NC004p, NE003s, NJ004p, NJ005p, NJ008p, NY004s, NY006p, OH008p, OR014p, PA004p, PA005p, PA008s, PA009p, PA010p, PA012p, SC001p, SC003p, TN007p, TN012p, VA001p, VA004p, VA005p, WI001p, WI009p, WI012p

Ceratophyllum demersum **Hornwort, common**

CO005p, IL002s, NC003p, PA004p, PA010p, WI001p, WI007p, WI012p

Cercis canadensis* var. *mexicana **Redbud, Mexican**

TX004p

Chamaecyparis thyoides **Cedar, Atlantic white**

CA003p, CT005p, DE002p, FL010p, FL014p, FL015p, LA002p, MA001b, MD002p, ME002p, MN006p, NC001p, NC007p, NJ005p, NJ008p, NY003p, NY004s, NY005p, OH008p, OR006p, OR008p, OR011p, OR014p, PA001p, PA012p, SC003p, VA004p, VA005p

Chamaedaphne calyculata **Leatherleaf**

MA004p, NJ005p, NY005p, OR014p, SC003p

Chasmanthium latifolium **Woodoats, Indian**

CA007p, CO003p, CO005p, CT002p, FL005p, FL010p, FL014p, FL016p, IL004b, IN001p, IN003p, KY002p, LA002p, MA004p, ME001s, MN009s, NC004p, NC005p, NC006p, NC008p, NE003s, NE011p, NJ001p, NY004s, OH002p, OH005p, OH007p, OH008p, OR004p, OR011p, OR014p, SC001p, SC003p, SC004s, TN013p, TN025b, TX004p, VA003p, VA005p, WI010p

Chasmanthium laxum **Woodoats, slender**

TN025p

Chelone glabra **Turtlehead, white**

CT002p, CT004p, IL004b, IL001s, IN003p, KY002b, MA002p, MA003p, MA004p, MN001p, MN009b, NC004p, NC005p, NE011p, NJ001p, NJ008p, NY002p, OR004p, OR011p, PA004p, PA008s, PA009p, PA010b, PA012p, SC001p, TN025p, VA003p, VA004p, VA005p, WI002p, WI009p, WI010p

Chelone lyonii **Turtlehead, pink**

CT002p, CT004p, KY003p, MA004p, MI001p, MN006p, MN010p, NC004p, NC005p, NE011p, NJ001p, NJ005p, NY002p, NY005p, OH007p, OH008p, OR004p, OR014p, SC001p, SC003p, TN013p, TN025p, UT002p, VA003p, WI010p

Chelone obliqua **Turtlehead, red**

CO005p, CT002p, GA001p, IL004p, MA004p, MN010p, NE011p, NJ007s, OH005b, OR014p, SC001p, TN025p, WI002p, WI010p

Chilopsis linearis **Desert willow**
AZ002p, CA002p, CA004s, CA005p, CA010s, MT001p, NE003s, NE005s, NY004s, OK004p, OR014p, SC003p, TX003p, TX004p, UT001s

Chrysobalanus icaco **Coco plum, Icaco**
FL005p, FL016p

Cicuta maculata **Water hemlock, spotted**
IL004b, MN009s

Cinna arundinacea **Woodreed, sweet (stout woodreed)**
IL004b, IN003p, MA003p, MN009b, NJ005p, NJ008p, PA008s, VA004p

Cladium mariscus ssp. jamaicense **Sawgrass, Jamaica**
FL005p, FL016p, FL017p, TX004p

Clematis baldwinii **Pine-hyacinth**
FL015p

Clematis crispa **Swamp leatherflower**
NC005p, NE003s, NY004s

Clematis ligusticifolia **Western white clematis**
CA002p, CA005p, CA007p, CA009p, CA017b, CO001p, CO003p, CO005p, ID002s, ID004s, MT001p, MT007p, NY004s, OR014p, WA003p

Clethra alnifolia **Sweetpepperbush, coast**
AR005p, CT001p, CT004p, CT005p, CT006p, DE002p, FL010p, FL012p, FL014p, FL015p, FL016p, IA004p, IL003p, IL005p, IN001p, LA002p, MA001s, MA003p, MA004p, MD002p, MD003p, ME002p, MI002p, MI004p, MN002p, MN006p, MN014p, NC002p, NC004p, NC005p, NC007p, NE003s, NJ001p, NJ004p, NJ005p, NJ006p, NJ008p, NJ009p, NY003p, NY004s, NY005p, NY006p, OH001b, OH002p, OH003p, OH008p, OH009p, OK001p, OR011p, OR014p, OR016p, PA001p, PA002p, PA004p, PA009p, PA010p, PA012p, SC001p, SC002p, SC003p, TN013p, TN014p, VA002p, VA004p, VA005p

Cliftonia monophylla **Buckwheat-tree**
FL014p, FL015p, LA002p, NY004s, OR014p, SC003p

Clusia minor **Cupey de monte**
FL004p

Coix lacryma-jobi **Tears, Job's**
NE003s

Colocasia esculenta **Coco yam (elephant's ear)**
AR002p, CT004p, FL012p, FL016p, GA001p, IA004p, MD001p, MS003p, NC003p, OH005p, PA002p, SC002p, SD003p

Comarum palustre **Purple marshlocks**
MN001p

Conocarpus erectus **Button mangrove**
FL002p, FL003p, FL004p, FL005p, FL006p, FL008p, FL016p

Conradina verticillata **Rosemary, Cumberland false**
MA004p, NC004p, NJ001p, OR014p, SC003p, WV001p

Coptis laciniata **Goldthread, Oregon**
OR002p

Coreopsis floridana **Tickseed, Florida**
FL014p, FL015p

Coreopsis integrifolia **Tickseed, fringeleaf**
AR005p, NC004p, SC003p

Coreopsis leavenworthii **Tickseed, Leavenworth's**
FL008p, FL012p, FL015p, FL016p

Coreopsis rosea **Tickseed, pink**
AR002p, CO001p, CO003p, CO005p, CT001p, CT002p, FL012p, FL016p, GA001p, IA004p, IL004p, IN001p, MA004p, MD003p, ME003s, MI001p, MI002p, MN002p, MN006p, MN010p, NE011p, NJ001p, NJ007s, NY002p, NY005p, OH002p, OH006p, OH010p, OR011p, OR014p, SC002p, SC003p, SC004s, TN013p, UT002p, VA003p, WI002p, WI010p, WI013p, WV001p

Cornus amomum **Dogwood, silky**
AR004p, AR005p, DE002p, FL010p, FL014p, IA001p, KY002p, MA001s, MA003p, MD002p, MI003b, MN002p, MN009b, MO002p, MT007p, NJ001p, NJ002p, NJ004p, NJ005p, NJ008p, NY003p, NY004s, NY006p, OH008p, PA004p, PA005p, PA006p, PA007p, PA008s, PA009p, PA010p, PA011p, PA012p, SC003p, TN003p, TN007p, TN008p, TN017p, TN021p, VA004p, VA005p, WI012p

Cornus asperifolia **Dogwood, toughleaf**
NY004s

Cornus foemina **Dogwood, stiff**
FL009p, FL014p, FL015p, MN004b, NY004s, VA005p

Cornus glabrata **Dogwood, brown**
CA002p, CA007p, NY004s

Cornus sericea* ssp. *sericea **Dogwood, redosier**
AR002p, AR004p, CA002p, CA005p, CA006p, CA007p, CA012p, CA017b, CO001p, CO003p, CO004p, CO005p, CT004p, CT005p, CT006p, DE001p, DE002p, IA001p, IA002p, IA004p, ID001p, ID002s, ID003p, IL006p, IN001p, MA001s, MA003p, MD002p, MD003p, MI002p, MI003p, MI004p, MN002p, MN003p, MN006p, MN009b, MN014p, MO002p, MT001p, MT002p, MT005p, MT007p, NC002p, NC005p, NJ002p, NJ005p, NJ008p, NY003p, NY004s, NY006p, OH003p, OH008p, OH009p, OK001p, OK004p, OK005p, OR005p, OR008p, OR009p, OR011p, OR014p, OR016p, PA001p, PA002p, PA004p, PA006p, PA007p, PA008s, PA009p, PA010p, PA011p, PA012p, PA013p, SD003p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN012p, TN016p, TN017p, TN018p, TN020p, TN021p, TN022p, TN024p, UT001s, UT002p, UT003p, UT004s, VA002p, VA004p, VA005p, WA003p, WI002s, WI003p, WI004p, WI012p, WY001s

Cosmos bipinnatus **Cosmos, garden**
AZ004s, CA004s, CA010s, CA011s, CO006s, CT008s, IA004s, ID004s, ME001s, ME003s, NE002s, NE003s, NJ007s, NY004s, OH005b, PA002s, PA008s, SC001p, SC004b, SD003s, TX002s, TX007s, UT001s, WI002s

Costus spicatus **Spiralflag, spiked**
FL012p

Cotula coronopifolia **Brassbuttons**
CA010s

Crataegus aestivalis **Hawthorn, May**
FL010p, FL012p, FL015p, LA005p, LA008s

Crataegus brachyacantha **Hawthorn, blueberry**
LA002p, LA008s, NY004s, OR014p, SC003p

Crataegus marshallii **Hawthorn, parsley**
FL015p, LA002p, LA008s, NY004s

Crataegus mollis **Hawthorn, Arnold**
LA008s, MA001s, MT006p, NY004s, OR014p, TN003p, VA005p

Crataegus opaca **Hawthorn, riverflat**
AR002p, FL001p, FL014p, LA002p, LA008s, OR014p

Crataegus spathulata **Hawthorn, littlehip**
FL015p, SC003p

Crataegus viridis **Hawthorn, green**
AL001p, CO005p, DE001p, DE002p, FL015p, IN001p, IN002p, KY001p, LA002p, LA008s, MN002p, MO002p, NJ004p, NY004s, OH003p, OH004p, OH008p, OR001p, OR003p, OR005p, OR009p, OR011p, OR014p, PA003p, TN003p, TN004p, TN009p, TN011p, TN012p, TN016p, TN018p, TN022p, TN024p, VA002p, WI003p

Crinum americanum **Swamplily, southern**
FL005p, FL015p, FL016p, FL017p, LA003p, MD001p, VA005p

Crossopetalum ilicifolium **Christmasberry**
FL004p

Crossopetalum rhacoma **Maidenberry**
FL002p

Ctenium aromaticum **Grass, toothache**
FL014p

Cuphea glutinosa **Waxweed, sticky**
FL012p

Cydista aequinoctialis **Guard withe**
FL016p

Cyperus eragrostis **Flatsedge, tall**
CA013p

Cyperus esculentus **Flatsedge, Chufa**
AL004p, AR003p, GA002s, GA003s, IL002s, MO011p, NY004s, PA008s, PA010b, WI001s

Cyperus haspan **Flatsedge, sheathed**
MD001p, NC003p, NE011p, SC001p

Cyperus involucratus **Umbrella plant**
CA004s, CT004p, FL012p, FL016p, LA007p, MD001p, NC003p, NE011p, SC001p

Cyperus odoratus **Flatsedge, fragrant**
FL016p, PA004p

Cyperus papyrus **Flatsedge, papyrus**
CA004s, CT004p, FL016p, LA003p, LA007p, MD001p, NC003p, SC001p

Cyperus strigosus **Flatsedge, strawcolored**
PA012p, WI012p

Cypripedium acaule **Moccasin flower (pink lady's slipper)**
KY002p

Cypripedium parviflorum **Lady's slipper, lesser yellow**
KY002p, NY005p

Cypripedium reginae **Lady's slipper, showy**
MN001p

Cyrilla racemiflora **Swamp titi (swamp cyrilla)**
FL001p, FL010p, FL014p, FL015p, LA008s, NC005p, NY004s, OR011p, OR014p, PA001p, PA004p, SC003p, VA005p

Cystopteris bulbifera **Bladderfern, bulblet**
IL004p, MN001p, MN009p, OR004p, OR014p

Dalea carnea **Whitetassels**
FL015p

Danthonia californica **Oatgrass, California**
CA013p, CA017b

Darlingtonia californica **Pitcherplant, California**
CA017p, NY001b

Darmera peltata **Indiana rhubarb**
OH008p, OR004p, WI010p

Decodon verticillatus **Swamp loosestrife**
CT009p, KY002p, MA004p, OH008p, PA004p

Decumaria barbara **Woodvamp**
AR005p, FL014p, NC004p, NC005p, NE011p, NY005p, OR014p, SC001p, SC003p

Delphinium glaucum **Larkspur, sierra**
CA007p

Deschampsia cespitosa **Hairgrass, tufted**
AR005p, CA002p, CA006p, CA007p, CA009p, CA010s, CA013b, CA017b, CO004p, CO005p, CO007s, CT002p, FL016p, ID002s, IL004b, MN002p, MN003p, MN006p, MN010p, MT004s, NC004p, NC008p, NE011p, NY005p, OH003p, OH008p, OR008p, OR009p, OR011p, OR014p, PA010p, UT001s, UT004s, WA002p, WA003p, WI010p, WY001s

Deschampsia danthonioides **Hairgrass, annual**
CA017b

Deschampsia elongata **Hairgrass, slender**
CA002p, CA010s

Dichanthelium acuminatum var. fasciculatum **Western panicgrass**
TX002s

Dichanthelium clandestinum **Deer-tongue**
GA003s, NJ003s, PA008s, PA010b, PA014s, PA015s

Dionaea muscipula **Venus flytrap**
CA004s, FL005p, NC004p, NJ007s, NY001b, PA010p

Dioscorea villosa **Yam, wild**
MA004p

Dipsacus fullonum* ssp. *sylvestris **Teasel, Fuller's**
IL001s, IL002s

Distichlis spicata **Saltgrass, inland**
AZ003s, CA002p, CA007p, CA009p, CA013p, CA017b, CO004p, FL005p, FL016p, FL017p, MA002p, MA003p, MD002p, NJ008p, NJ009p, UT001s, UT004s, VA001p, VA004p

Dodecatheon jeffreyi **Shootingstar, Sierra**
ID002s, NY005p, OR014p

Dodecatheon pulchellum **Shootingstar, darkthroat**
CO005p, ID002s, ID004s, MN009s

Doellingeria umbellata **Parasol whitetop**
IL004b, IN003p, MN001p, MN004b, MN009b, NC005p, PA008s

Dracopis amplexicaulis **Coneflower, clasping**
CA010s, CA011s, CO006s, IA003s, IL001s, NE002s, OK002s, TN025s, TX002s, TX007s, UT001s

Drosera capillaris **Sundew, pink**
NY001p

Drosera filiformis **Sundew, threadleaf**
NJ001p, NY001b

Drosera intermedia **Sundew, spoonleaf**
NJ001p, NY001b

Drosera rotundifolia **Sundew, roundleaf**
NC004p, NY001b, PA010p

Dryopteris carthusiana **Woodfern, spinulose**
MN001p, MN009p, NJ001p, NY002p, NY005p, OH008p, OR014p, SC002p, TN007p, TN015p, TN019p, TN022p, TN023p, VA005p, WI010p

Dryopteris celsa **Fern, log**
AR002p, CT002p, FL012p, FL016p, NC005p, NJ001p, NY005p, OH005p, OR014p, SC001p, SC003p, TN013p, WA001p

Dryopteris clintoniana **Woodfern, Clinton's**
CT002p, NY005p

Dryopteris cristata **Woodfern, crested**
NJ001p

Dryopteris expansa **Woodfern, spreading**
AR002p, CO003p, CT002p, FL012p, NY005p, OH005p, OR004p, OR011p, VA003p, WA001p

Dryopteris ludoviciana **Woodfern, southern**
SC003p

Dryopteris X boottii **Woodfern, Boott's**
CT002p, NY005p

Dulichium arundinaceum **Sedge, threeway**
KY002p, MA003p, MA004p, MD001p, NC003p, PA008s

Echinochloa crus-galli **Barnyardgrass**
GA002s, IL002s, NJ003s, PA008s, PA010b, WI001s, TX002s

Echinocystis lobata **Cucumber, wild**
MN009s

Echinodorus cordifolius **Burrhead, creeping**
MS006p, MS007p, MS008p, MS009p, TN026p

Egeria densa **Waterweed, Brazilian**
NC003p

Eichhornia crassipes **Water hyacinth, common**
CT004p, MD001p, NC003p

Elaeagnus angustifolia **Olive, Russian**
AR002p, CA004s, CA013p, CO001p, CO003p, CO005p, CO008p, DE002p, GA002p, IA002p, IA004p, ID003p, ID002s, IL003p, IL005p, IL006p, LA005p, LA008s, MA001s, MI003b, MN002p, MN014p, MN020p, MO002p, MT002p, MT005p, MT006p, MT007p, MT015p, NE003s, NE005s, NJ004p, NV001p, NY004s, NY006p, OH008p, OH010p, OK001p, OK004p, OK007p, OR005p, OR009p, OR011p, OR014p, OR015p, PA005p, PA006p, PA007p, SD003p, TN003p, TN004p, TN006p, TN007p, TN008p, TN017p, TN018p, TN019p, TN020p, TX003p, UT002p, UT004s, VA005p, WA003p, WI002s, WY001s

Eleocharis acicularis **Spikerush, needle (least spikerush)**
CA003p, CA009p, CO004p, CO005p, MN009s, MT001p, NJ005p, PA004p, PA010p, WI007p, WI012b

Eleocharis baldwinii **Spikerush, Baldwin's**
VA005p

Eleocharis cellulosa **Spikerush, gulf coast**
FL017p

Eleocharis dulcis **Chinese waterchestnut**
MD001p, NC008p

Eleocharis interstincta **Spikerush, knotted**
FL017p

Eleocharis montevidensis **Spikerush, sand**
MD001p, NC003p, NJ006p

Eleocharis obtusa **Spikerush, blunt**
CT009p, MA002p, MD002p, NJ005p, PA004p, PA008s, PA009p, PA010p, WI012b

Eleocharis olivacea **Spikerush, bright green**
NJ005p

Eleocharis palustris **Spikerush, creeping**
CA002p, CA007p, CA009p, CA013p, CA017b, CO004p, CO005p, IN003p, MA003P, MT001p, OR008p, OR009p, PA010p, PA012p, WI001p, WI007p, WI012b

Eleocharis parvula **Spikerush, dwarf**
PA010p, UT001s

Eleocharis quadrangulata **Spikerush, squarestem**
MD001p

Eleocharis radicans **Spikerush, rooted**
CA003p

Eleocharis vivipara **Spikerush, viviparous**
NC003p

Elodea canadensis **Waterweed, Canadian**
CT004p, NC003p, NC008p, PA004p, PA010p, WI001p, WI007p, WI012p

Elymus riparius **Wildrye, riverbank**
IL004b, MN009b, NJ005p, NJ008p, NY004s, PA008s, VA004p

Elymus virginicus **Wildrye, Virginia**
AR004p, AR005p, IA003s, IA005s, IL004b, KY002p, MN004b, MN009b, MN013s, MO001s, NE002s, NE007s, NJ005p, NJ008p, OK002s, PA008s, PA010b, VA004p, WI008s, WI009s

Empetrum nigrum **Crowberry, black**
OR014p

Epilobium coloratum **Willowherb, purpleleaf**
MN004b, PA008s

Epipactis gigantea **Helloborine, giant**
CA002p, CA013p

Equisetum arvense **Horsetail, field**
AR005p, CA012p, CA017p, PA010p, WI001p, WI007p, WI012p

Equisetum fluviatile **Horsetail, water**
PA010p, WI001p, WI012p

Equisetum hyemale **Horsetail, rough**
CA013p, FL012p, KY002p, LA003p, LA007p, MA004p, MD001p, NC003p, NC004p, NC005p, NC008p, NE011p, OH008p, OR014p, PA010p, TN013p, WA001p

Equisetum sylvaticum **Horsetail, woodland**
WI012p

Equisetum telmateia **Horsetail, giant**
CA013p

Eragrostis campestris **Lovegrass, coastal**
FL009p

Erigeron peregrinus **Fleabane, subalpine**
MT001p

Erigeron philadelphicus **Fleabane, Philadelphia**
CA002p

Erigeron vernus **Fleabane, early whitetop**
NC005p

Eriophorum angustifolium **Cottongrass, tall**
OH008p

Eriophorum virginicum **Cottongrass, tawny**
PA008s, PA010p

Eryngium aquaticum **Button snakeroot**
TN002p

Eryngium yuccifolium **Rattlesnakemaster**
CT002p, FL015p, IA003s, IL001s, IL004b, IN003p, KY002b, KY003p, MI001p, MN004b, MN009b, MN011s, MO003b, NC004p, NC005p, NE003s, NE011p, NJ001p, OH008p, OR014p, TN025b, WI006p, WI008b, WI009b

Euonymus americana **Strawberry bush**
AR002p, CT005p, FL012p, FL015p, KY002p, LA002p, LA008s, MA001s, MD001p, NC004p, NC005p, NJ001p, NY004s, OR014p, SC003p, TN003p, TN007p, TN013p, TN017p, TN019p, VA005p

Eupatorium coelestinum **Mistflower**
FL015p, KY002b, LA002p, MN009s, MN010p, NC004p, NC005p, NE011p, NJ001p, OH002p, OH005p, OH007p, OR014p, TN013p, TN025b, WI010p

Eupatorium dubium **Joepyeweed, coastalplain**
MD002p, NJ005p

Eupatorium fistulosum **Trumpetweed**
CT004p, FL014p, IA003s, IN003p, KY003p, NC004p, NJ005p, NY004s, NY005p, OH007p, OH008p, PA008s, PA009p, PA012p, SC001p, SC002p, TN025b, VA002p, VA003p, WI010p

Eupatorium maculatum* var. *maculatum **Joepyeweed, spotted**
AR005p, CT002p, CT009b, IL001s, IL004p, IN003p, KY002b, OH008p, MA003p, MN002p, MN004s, MN006p, MN007s, MN009b, MN010p, MN011s, NC005p, NE003s, NJ001p, NJ007s, NY004s, NY005p, OR004p, OR014p, PA008s, PA010b, SC001p, TN013p, WI007p, WI008b, WI009b, WI010p, WI012b, WI013p

Eupatorium perfoliatum **Boneset, common**
CT009b, IL001s, IL004p, IN003p, KY002p, KY003b, ME001s, MA002p, MA004p, MD002p, MN004b, MN007s, MN009b, MN011s, NC005p, NJ001p, NJ008p, PA004p, PA008s, PA009p, PA010b, PA012p, TN025p, VA004p, VA005p, WI006p, WI007p, WI008b, WI009b, WI012b

Eupatorium resinosum **Thoroughwort, pine barrens**
NJ005p

Eustoma exaltatum **Prairie gentian, catchfly**
FL015p

Eustoma russellianum **Prairie gentian, showy**
TX004p

Euthamia graminifolia **Goldentop, flat-top**
MN004b, PA008s, PA009p

Festuca rubra **Fescue, red**
AR003p, AZ001s, CA004s, CA006p, CA007p, CA010s, CA013b, CA014s, CA015s, CA016s, CO007s, GA003s, IA003s, ID004s, ID006s, MT003s, MT004s, NE002s, NJ003s, NM001s, PA008s, SD001s, SD002s, UT001s, UT004s, UT006s, WY001s

Ficus aurea **Fig, Florida strangler**
FL002p, FL004p, FL006p, FL008p

Filipendula rubra **Queen of the prairie**
CO003p, CO005p, CT002p, CT004p, GA001p, IA004p, IL004p, IN003p, MN002p, MN006p, MN009p, MN010p, NE011p, NY002p, OH007p, OR011p, OR012p, OR014p, SC001p, SC002p, WI002p, WI006p, WI008b, WI009p, WI010p, WI013p

Fimbristylis castanea **Fimbry, marsh**
FL017p

Flaveria linearis **Flaveria, narrowleaf yellowtop**
FL006p

Forestiera acuminata **Swampprivet**
FL015p, OR014p, SC003p

Fothergilla gardenii **Witchalder, dwarf**
CT001p, CT004p, CT005p, CT006p, FL015p, IN001p, LA001p, LA002p, MI002p, MI004p, MN002p, MN006p, NC002p, NC005p, NJ001p, NY003p, NY005p, NY006p, OH002p, OH003p, OH008p, OK001p, OR005p, OR006p, OR011p, OR014p, PA001p, SC001p, SC002p, SC003p, TN004p, TN013p, TN014p, TN016p, VA002p, VA005p

Frankenia salina **Seaheath, alkali**
CA002p

Fraxinus caroliniana **Ash, Carolina**
FL001p, FL003p, FL009p, FL016p, LA008s

Fraxinus latifolia **Ash, Oregon**
CA004s, CA007p, CA008s, CA009p, CA017b, ID002s, MT001p, MT007p, OR008p, OR009p, OR014p

Fraxinus nigra **Ash, black**
MA003p, MN002p, MN006p, MN009p, MN012p, MT005p, MT006p, MT007p, NY004s

Fraxinus pennsylvanica **Ash, green**
AL006p, AR001p, AR002p, CA004s, CO001p, CO003p, CO005p, CT001p, DE001p, DE002p, FL001p, FL005p, FL007p, FL010p, FL012p, FL013p, FL014p, FL016p, GA002p, IA001p, IA004p, ID002s, ID003p, ID007p, IN001p, IN002p, KY001p, LA001p, LA003p, LA004p, LA005p, LA008s, MA001s, MA003p, MD002p, MI002p, MI003b, MI004p, MN002p, MN003p, MN005p, MN006p, MN012p, MN014p, MO002p, MS001p, MS004p, MT002p, MT005p, MT006p, MT007p, MT015p, NC001p, NJ004p, NJ005p, NJ008p, NV001p, NY003p, NY004s, NY006p, NV001p, OH003p, OH004p, OH008p, OK001p, OK004p, OK007p, OR001p, OR003p, OR005p, OR008p, OR009p, OR015p, PA003p, PA005p, PA007p, PA008s, PA009p, PA010p, PA011p, PA012p, SD003p, TN001p, TN002p, TN003p, TN004p, TN006p, TN007p, TN008p, TN009p, TN010p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, TX003p, TX008p, TX013p, TX014p, TX015p, UT002p, UT003p, VA002p, VA004p, VA005p, WI003p, WI012p

Fraxinus profunda **Ash, pumpkin**
FL014p, FL015p, LA008s

Fraxinus velutina **Ash, velvet**
AR002p, CA004s, LA008s, NY004s, OK001p

Gelsemium rankinii **Trumpetflower, Rankin's**
FL012p, NC004p, NY005p, SC001p, SC003p

Gentiana andrewsii **Gentian, closed bottle**
CT004p, IL001s, IL004b, IN003p, KY002p, MN004b, MN001p, MN009b, MN011s, NC005p, NE003s, NE011p, NJ001p, NJ005p, OR004p, TN006p, TN013p, WI006p, WI008b, WI010p

Gentiana calycosa **Gentian, Rainier pleated**
ID002s

Gentiana clausa **Gentian, bottle**
NJ005p, PA008s

Gentiana saponaria **Harvestbells**
SC003p

Geum aleppicum **Avens, yellow**
MN009s, PA008s

Geum laciniatum **Avens, rough**
PA008s

Geum macrophyllum **Avens, largeleaf**
CO005p, ID004s

Geum rivale **Avens, purple**
MA004p, MN010p, OR014p

Glandularia tampensis **Mock vervain, Tampa**
FL015p

Gleditsia aquatica **Water locust**
MO002p, NY004s

Glyceria canadensis **Rattlesnake mannagrass**
CT009b, MA002p, MA003p, MN009s, PA008s, PA009p, PA012p, WI008s

Glyceria elata **Fowl mannagrass**
CA017b, MT001p, OR009p

Glyceria grandis **American mannagrass**
CO004p, MN009s, MN011s, MT001p, PA004p, PA008s, WI008s

Glyceria maxima **Reed mannagrass**
CT002p, MI001p, MN010p, OH007p, OH008p, OR011p

Glyceria melicaria **Melic mannagrass**
PA008s

Glyceria obtusa **Atlantic mannagrass**
NJ005p

Glyceria occidentalis **Northwestern mannagrass**
CA017b, MT001p, OR009p, PA008s, UT001s

Glyceria striata **Fowl mannagrass**
CO004p, CO005p, IL004b, IN003p, KY002p, MA002p, MN009s, NJ008p, PA008s, PA009p, PA010b, PA012p, VA004p, WI008s, WI009s, WI012b

Gordonia lasianthus **Bay, loblolly**
FL001p, FL003p, FL005p, FL007p, FL009p, FL010p, FL012p, FL013p, FL014p, FL015p, FL016p, LA001p, LA003p, NY004s, SC003p, VA005p

Gratiola aurea **Hedgehyssop, golden**
NJ005p

Grindelia hirsutula var. hirsutula **Hairy gumweed**
CA009p

Grindelia stricta* var. *angustifolia **Oregon gumweed**
CA009p

Habranthus tubispathus **Rio Grande copperlily**
AR005p, SC003p

Hamamelis vernalis **Witchhazel, Ozark**
AR005p, CT006p, FL015p, IN001p, IN002p, MA001s, ME002p, MI004p, MN002p, MO005p, MT007p, NC005p, NJ001p, NJ004p, NJ005p, NY003p, NY004s, NY005p, OH003p, OH008p, OR005p, OR011p, OR014p, PA001p, SC003p, TN004p, TN016p, VA005p

Hedychium coronarium **White garland-lily**
AR002p, FL012p, LA003p, LA007p, MS003p, NC004p, NC007p, SC003p

Heimia salicifolia **Shrubby yellowcrest**
OR014p

Helenium autumnale **Sneezeweed, common**
CO003p, CO005p, CT003p, IL001s, IL004p, IN003p, KY002b, ME001s, MN002p, MN004b, MN007s, MN009b, MN011s, NC004p, NE003s, NJ007s, NY004s, OH005s, OR011p, PA008s, SC001p, SC004s, VA003p, WI005p, WI008b, WI010p

Helenium bigelovii **Sneezeweed, Bigelow's**
CA007p, OR014p

Helenium flexuosum **Sneezeweed, purplehead**
MN009s, NC005p

Helenium puberulum **Rosilla**
CA002p

Helianthus agrestis **Sunflower, southeastern**
FL015p

Helianthus angustifolius **Sunflower, swamp**
AR004p, AR005p, CT002p, FL012p, FL014p, FL015p, KY002b, LA002p, NC004p, NC005p, OH007p, OH008p, SC001p

Helianthus giganteus **Sunflower, tall**
MN001p, MN004b, NC004p

Helianthus grosseserratus **Sunflower, sawtooth**
AR005p, MN009s, WI008b, WI009s

Helianthus heterophyllus **Sunflower, variableleaf**
FL014p

Helianthus nuttallii **Sunflower, Nuttall's**
AR005p, CO004p, CO005p, PA010p

Helianthus simulans **Sunflower, muck**
TN013p, TN025p

Heracleum maximum **Cowparsnip, common**
CA013p, MT001p, WI008s

Heteranthera dubia **Mudplantain, grassleaf**
PA010p

Hibiscus coccineus **Rosemallow, scarlet**
AR005p, FL010p, FL012p, FL015p, FL016p, LA002p, MA004p, NC005p, NY005p, OR014p, SC001p, SC003p, TN025p

Hibiscus dasycalyx **Rosemallow, Neches River**
SC003p

Hibiscus grandiflorus **Rosemallow, swamp**
FL010p, FL012p, FL015p, FL016p, SC003p, TN025p

Hibiscus laevis **Rosemallow, halberdleaf**
FL012p, MN009b, NC005p, NE003s, SC003p, TN025p

Hibiscus moscheutos **Rosemallow, crimsoneyed**
AR002p, CO003p, CO005p, CT002p, CT003p, FL012p, FL013p, FL015p, GA001p, IL004p, IL005p, IL006p, IN003p, KY002b, LA003p, MA001s, MA004p, MD002p, MI001p, MI005p, MN002p, MN010p, MN014p, MN020p, NC004p, NC005p, NE003s, NE011p, NJ007s, NJ008p, NY002p, NY004s, OH003p, OH005b, OH007p, OH008p, OK001p, OR011p, OR014p, PA004p, PA008s, PA009p, PA010b, PA012p, SC001p, SC002p, SC003p, SC004p, TN006p, TN013p, TN017p, TN025p, UT002p, VA001p, VA004p, VA005p, WI002b

Hibiscus tiliaceus **Rosemallow, sea**
CA004s, FL016p

Hierochloe odorata **Grass, vanilla**
MA004p, MN009b, NE003s, NE011p, WI006p, WI008b

Hoita macrostachya **Leather-root, large**
CA002p

Hoita orbicularis **Leather-root, roundleaf**
CA002p

Hordeum brachyantherum **Barley, meadow**
CA002p, CA004s, CA007p, CA010s, CA013s, UT001s

Hordeum jubatum **Barley, foxtail**
CA004s, PA008s

Houstonia serpyllifolia **Bluet, thymeleaf**
NC005p

Huperzia lucidula **Clubmoss, shining**
NC005p

Hydrocleys nymphoides **Waterpoppy**
MD001p, NC003p, NC008p

Hydrocotyle ranunculoides **Marshpennywort, floating**
CA017p

Hydrocotyle verticillata **Marshpennywort, whorled**
MD001p

Hydrolea corymbosa **Skyflower**
FL015p

Hydrophyllum virginianum **Waterleaf, Virginia Shawnee salad**
IL004p, MN009p, NC005p, TN025p, WI008b

Hymenocallis caroliniana **Spiderlily, Carolina**
LA007p

Hymenocallis floridana **Spiderlily, Florida**
FL015p

Hymenocallis latifolia **Spiderlily, perfumed**
FL002p, FL003p, FL004p, FL008p, FL012p, FL016p

Hymenocallis liriosome **Spring spiderlily**
CT004p

Hypericum canadense **St. Johnswort, Canadian**
PA004p

Hypericum densiflorum **St. Johnswort, bushy**
MA001s, SC003p

Hypericum fasciculatum **St. Johnswort, sandweed peelbark**
FL015p

Hypericum galioides **St. John'swort, bedstraw**
NC004p

Hypericum kalmianum **St. Johnswort, Kalm's**
CO001p, CO005p, CT004p, IA002p, IL004b, MA001s, MN002p, MN006p, MN009p, MN020p, OH008p, OH009p, OR005p, OR011p, OR014p, WI008s

Hypericum lissophloeus **St. Johnswort, smoothbark**
SC003p

Hypericum myrtifolium **St. Johnswort, myrtleleaf**
FL015p

Hypericum nudiflorum **St. Johnswort, early**
SC003p

Hypoxis hirsuta **Common goldstar**
MN009b, NC005p, NJ001p, OR004p, TN010p

Ilex amelanchier **Holly, Sarvis**
FL015p, SC003p

Ilex cassine **Holly, Dahoon**
AR002p, FL001p, FL003p, FL004p, FL005p, FL007p, FL008p, FL009p, FL010p, FL014p, FL015p, FL016p, LA002p, LA008s, SC003p

Ilex coriacea **Holly, large gallberry**
FL015p

Ilex decidua **Holly, possumhaw**
AL001p, AR002p, FL010p, FL014p, FL015p, KY002p, LA002p, LA008s, MA001s, MN002p, NY004s, NY005p, OH003p, OH008p, OK001p, OR002p, OR011p, OR014p, PA012p, SC001p, SC003p, TN004p, VA005p

Ilex glabra **Inkberry**
AL001p, AR005p, CT001p, CT004p, CT005p, CT006p, DE001p, FL001p, FL005p, FL010p, FL016p, IN001p, LA002p, LA008s, MA001s, MD003p, MI002p, MI004p, MN002p, MN006p, NC002p, NJ001p, NJ004p, NJ006p, NJ008p, NY003p, NY004s, NY005p, NY006p, OH001p, OH003p, OH008p, OH009p, OK001p, OR008p, OR011p, PA001p, PA004p, PA009p, PA012p, SC001p, SC003p, TN004p, TN014p, TN016p, VA002p, VA004p, VA005p

Ilex laevigata **Holly, smooth winterberry**
OR014p

Ilex longipes **Holly, Georgia**
FL015p

Ilex myrtifolia **Holly, myrtle dehoon**
FL001p, FL009p, FL010p, FL014p, FL015p, OR014p, SC003p

Ilex verticillata **Winterberry, common**
AL001p, AR002p, AR005p, CT001p, CT004p, CT005p, CT006p, DE001p, DE002p, FL010p, FL015p, FL016p, ID002s, IN001p, KY002p, MA001s, MA003p, MD002p, MD003p, ME002p, MI004p, MI005p, MN002p, MN004b, MN006p, MN009p, NC002p, NJ001p, OH001p, OR014p, NJ004p, NJ005p, NJ008p, NY003p, NY004s, NY005p, NY006p, OH001p, OH003p, OH008p, OH009p, OK001p, OR005p, OR011p, PA001p, PA005p, PA008s, PA009p, PA010p, PA011p, PA012p, SC001p, SC002p, SC003p, TN003p, TN004p, TN007p, TN012p, TN013p, TN014p, TN016p, TN018p, TN022p, TN024p, VA002p, VA004p, VA005p, WI003p, WI004p

Illicium floridanum **Anisetree, Florida**
FL005p, FL012p, FL014p, FL015p, FL016p, LA002p, LA003p, LA007p, NC004p, NC007p, NY005p, OR014p, SC001p, SC003p, TN013p, VA005p

Illicium parviflorum **Anisetree, yellow**
FL010p, FL012p, FL014p, FL015p, FL016p, LA002p, NC004p, OR014p, SC001p, SC003p, TN014p, VA005p

Impatiens capensis **Jewelweed (touch-me-not, spotted)**
IL001s, MA004p, MN009s, PA004p, PA008s, PA010p

Impatiens pallida **Touch-me-not, pale**
MN009s, PA008s, PA010p

Ipomoea alba **Morning-glory, tropical white**
CT008s, SC004s

Ipomoea lacunosa — **Whitestar**
IL002s

Iris brevicaulis — **Iris, zigzag**
LA002p, MN008p, SC003p

Iris fulva — **Iris, copper**
KY002p, LA002p, MN008p, NC004p, NC005p, OR014p, SC003p, TN013p, TN025p

Iris hexagona — **Iris, Dixie**
FL015p, FL016p, SC003p

Iris missouriensis — **Iris, Rocky Mountain**
CA002p, CA007p, CA009p, CA017p, CO001p, CO002s, CO004p, CO005p, CO006s, ID002s, ID004s, MN008p, MN009s, MT001p, OR014p, OR016p, PA010p, UT001s, UT002p, WY001s

Iris prismatica — **Iris, slender blue**
AR005p, CT009p, MN009b, NC005p, NJ001p, OR004p, OR014p, SC001p

Iris pseudacorus — **Iris, paleyellow**
CA017b, CO003p, CO004p, CO005p, CT002p, CT004p, CT009p, DE002p, FL012p, GA001p, IL004p, IN001p, LA003p, LA007p, MA004p, MD001p, MD002p, MI001p, MN006p, MN008p, MN010p, MS003p, MT001p, NC003p, NC004p, NC005p, NC008p, NE003s, NE011p, NJ001p, NJ005p, NJ006p, NJ008p, NY002p, NY004s, NY005p, OH003p, OH008p, OR004p, OR008p, OR009p, OR011p, OR014p, PA004p, PA008s, PA009p, PA010b, PA012p, SC001p, SC003p, TN025p, VA001p, VA002p, VA003p, VA004p, VA005p, WI001p, WI007p, WI010p, WI011p, WI012b

Iris tridentata — **Iris, savannah**
NC003p

Iris versicolor — **Blueflag, harlequin**
CT002p, CT004p, CT009b, DE002p, IN003p, KY002p, MA002p, MA003p, MA004p, MD001p, MD002p, MN001p, MN004b, MN006p, MN007s, MN008p, MN009b, MN010p, MT001p, NC003p, NC004p, NC005p, NE003s, NE011p, NJ001p, NJ005p, NJ008p, NY002p, OH008p, OR004p, OR011p, OR014p, PA004p, PA008s, PA009p, PA010p, PA012p, TN013p, TN025p, VA001p, VA003p, VA004p, VA005p, WI001p, WI007p, WI009b, WI012b

Iris virginica — **Iris, Virginia (blueflag iris)**
FL005p, FL009p, FL010p, FL012p, FL016p, FL017p, IL004b, IN001p, IN003p, LA002p, LA007p, MN008p, MN009b, MN011s, NC004p, NC005p, NC008p, TX004p

Isachne confusa — **Isachne**
FL003p

Itea virginica **Virginia sweetspire (Virginia willow)**
AR005p, CT001p, CT004p, CT005p, DE002p, FL001p, FL003p, FL005p, FL010p, FL012p, FL014p, FL015p, FL016p, FL017p, IN001p, KY002p, LA002p, LA007p, MA001s, MA004p, MD002p, MD003p, MI002p, MN002p, MO005p, NC002p, NC004p, NC005p, NC007p, NJ001p, NJ004p, NJ005p, NJ008p, NY004s, NY005p, NY006p, OH003p, OH008p, OK001p, OR002p, OR005p, OR008p, OR011p, OR014p, PA001p, PA009p, PA012p, SC001p, SC003p, TN007p, TN012p, TN013p, TN014p, TN016p, VA002p, VA004p, VA005p

Iva frutescens **Jesuit's bark (marshelder)**
FL005p, MD001p, NJ009p, NJ008p, PA009p, VA001p, VA004p, VA005p

Iva hayesiana **San Diego povertyweed**
CA002p, CA005p, CA010s

Iva imbricata **Seacoast marshelder**
FL006p, FL008p, FL016p

Jacquinia keyensis **Joewood**
FL002p, FL004p, FL008p

Juglans major **Walnut, Arizona**
NY004s

Juncus acuminatus **Rush, tapertip**
CT009s, NJ005p

Juncus acutus **Rush, spiny**
CA002p, CA010s

Juncus arcticus **Rush, arctic**
CO005p

Juncus articulatus **Rush, jointedleaf**
CT009b

Juncus balticus **Rush, Baltic**
CA007p, CA009p, CA013p, CA017b, CO004p, MT001p, OR009p, PA010p, UT001s, WI012b

Juncus bolanderi **Rush, Bolander's**
CA017b

Juncus brachycarpus **Rush, whiteroot**
MN009s

Juncus brevicaudatus **Rush, narrowpanicle**
CT009b

Juncus bufonius **Rush, toad**
CA017b

Juncus caesariensis **Rush, New Jersey**
NJ005p

Juncus canadensis **Rush, Canadian**
CT009b, MA002p, MA003p, NJ005p, NJ008p, PA008s, PA010p, VA004p, WI001p, WI012p

Juncus compressus **Rush, roundfruit**
CO003p

Juncus confusus **Rush, Colorado**
CO004p, CO005p, MT001p, PA010p

Juncus dichotomus **Rush, forked**
NJ005p

Juncus drummondii **Rush, Drummond's**
CO004p, CO005p, MT001p

Juncus dubius **Rush, dubius**
CA002p

Juncus dudleyi **Rush, Dudley's**
WI008s

Juncus effusus **Rush, common (soft rush)**
AR005p, CA002p, CA003p, CA006p, CA007p, CA013p, CA017b, CT002p, CT009b, FL005p, FL009p, FL010p, FL012p, FL016p, FL017p, ID002s, IN003p, KY002p, LA002p, MA002p, MA003p, MA004p, MD002p, MN009s, MS003p, MT001p, NC003p, NC004p, NC008p, NE011p, NJ001p, NJ005p, NJ008p, NY005p, OH008p, OR008p, OR009p, OR014p, PA004p, PA008s, PA009p, PA010b, PA012p, SC001p, VA001p, VA004p, VA005p, WI001p, WI007p, WI008s, WI012b

Juncus ensifolius **Rush, swordleaf**
AR005p, MT001p, NC005p, OH008p, UT003p

Juncus gerardii **Rush, saltmeadow**
MA002p, NJ008p, VA004p

Juncus inflexus **Rush, European meadow**
CA006p

Juncus interior **Rush, inland**
MN009s

Juncus longistylis **Rush, longstyle**
CO004p, PA010p

Juncus macrophyllus **Rush, longleaf**
CA002p

Juncus marginatus **Rush, grassleaf**
CT009b, NJ005p

Juncus mertensianus **Rush, Mertens'**
CO004p, CO005p, MT001p, PA010p

Juncus mexicanus **Rush, Mexican**
CA007p

Juncus militaris **Rush, bayonet**
CT009p, NJ008p, VA004p

Juncus nevadensis **Rush, Sierra**
CA017b, UT003p

Juncus nodosus **Rush, knotted**
CO004p, CO005p, MA002p, MN009s, MT001p, PA010p

Juncus patens **Rush, spreading**
CA002p, CA006p, CA007p, CA007p, CA009p, CA013p, CA017b, NC004p, OR009p

Juncus phaeocephalus **Rush, brownhead**
CA002p

Juncus roemerianus **Rush, needlegrass**
FL005p, FL016p, FL017p, MD002p, NJ008p, VA004p

Juncus saximontanus **Rush, Rocky Mountain**
CO004p, PA010p

Juncus scirpoides **Rush, needlepod**
NJ005p

Juncus tenuis **Rush, poverty (path rush)**
CA017b, CO004p, CO005p, MN009b, NE011p, OR009p, PA008s, PA010p, WI008s, WI012b

Juncus torreyi **Rush, Torrey's**
CO003p, CO004p, CO005p, IN003p, MN009b, MT001p, PA008s, PA010p, WI001p, WI007p, WI008s, WI012p

Juncus triglumis **Rush, threehulled**
CO004p, CO005p, PA010p

Juncus xiphioides **Rush, irisleaf**
AR005p, CA007p, CA009p

Juniperus virginiana var. silicicola **Cedar, southern red**
CA004s, FL003p, FL005p, FL008p, FL009p, FL010p, FL014p, FL015p, FL016p, NY004s, SC001p

Justicia americana **Water-willow, American**
PA010p

Justicia ovata **Water-willow, looseflower**
FL012p

Kalmia cuneata **Whitewicky**
NY005p

Kalmia hirsuta **Laurel, hairy**
SC003p

Kalmia microphylla **Laurel, alpine**
OR002p

Kalmia polifolia **Laurel, bog**
MN001p, OR014p

Kosteletzkya virginica **Mallow, Virginia saltmarsh**
FL015p, LA002p, MD002p, NC004p, TN013p, TN025p, VA001p

Laguncularia racemosa **Mangrove, white**
FL002p, FL005p, FL008p, FL016p

Laportea canadensis **Woodnettle, Canadian**
MA004p

Larix laricina **Tamarack**
CT005p, MA001s, MA003p, MN001p, MN002p, MN003p, MN004b, MN009p, MT007p, NY004s, NY005p, OR006p, OR011p, OR014p, PA010p, PA012p

Lasthenia glabrata **Goldfields, yellowray**
CA010s, CA011s, CO006s

Ledum glandulosum **Labrador tea, western**
OR011p

Ledum groenlandicum **Labrador tea, bog**
CT004p, MN001p, NY005p, OR014p

Leersia oryzoides **Cutgrass, rice**
IL004s, IN003p, MA002p, MA003p, MD002p, MN009s, NJ005p, NJ008p, PA004p, PA008s, PA009p, PA010b, PA012p, VA001p, VA004p, VA005p, WI001p, WI007p, WI008s, WI012b

Leersia virginica **Whitegrass**
IL004p, NJ008p, PA008s, VA004p

Leitneria floridana **Corkwood**
FL014p, FL015p, NY004s, SC003p

Lemna minor **Duckweed, lesser**
CA007p, CA017p, NC003p, OR009p, WI001p, WI012p

Lemna trisulca **Duckweed, star**
WI001p, WI012p

Leucothoe axillaris **Doghobble, coastal**
AR002p, CT001p, CT004p, CT005p, FL015p, IN001p, MA001s, MD003p, NJ004p, NY004s, NY005p, OH003p, OH008p, OK001p, OR008p, OR009p, OR011p, OR014p, PA001p

Leucothoe davisiae **Sierra laurel**
OR014p

Leucothoe racemosa **Swamp doghobble**
FL015p, MA001s, MD002p, NC005p, NJ005p, NY005p, SC003p

Liatris lancifolia **Lanceleaf blazing star**
MN009s

Lilium catesbaei **Lily, pine**
FL015p

Lilium kelleyanum **Lily, Kelley's**
CA002p

Lilium michiganense **Lily, Michigan**
MN004p, MN009b

Lilium pardalinum **Lily, leopard**
CA002p, CA007p, CA013p, CA017b, OR014p

Lilium pardalinum* ssp. *wigginsii **Lily, Wiggin's**
CA002p

Lilium parryi **Lily, lemon**
CA002p

Lilium philadelphicum **Lily, wood**
MN009b

Lilium superbum **Lily, turk's-cap**
CT004p, MA004p, NC004p, NE011p, NJ001p, NY004s, OR004p, PA008s, PA010p, TN003p, TN010p, TN013p, TN015p, TN017p, TN019p, TN022p, TN023p, TN025p, WI006p, WI008b, WI009p, WI012p

Limnanthes douglasii **Meadowfoam, Douglas'**
NE003s

Limonium californicum **Sealavender, California**
CA005p, CA009p, CA010s

Limonium sinuatum **Sealavender, wavyleaf**
CA010s, CA011s, CT008s, ME001s, ME003s, MT015p, NJ007s, NY004s, OH005b, PA002s, SC004s, WI002p

Lindera benzoin **Spicebush, northern**
AR005p, CT005p, DE002p, FL015p, IL001s, KY002p, LA008s, MA001s, MA003p, MD002p, MN002p, MN009p, NC001p, NC005p, NJ001p, NJ004p, NJ008p, NY003p, NY004s, NY005p, OH008p, OR011p, OR014p, OR016p, PA001p, PA004p, PA008s, PA009p, PA010p, PA012p, SC003p, TN003p, TN005p, TN007p, TN010p, TN018p, TN017p, TN022p, VA004p, VA005p, WI012p

Lindera subcoriacea **Spicebush, bog**
SC003p

Lindernia grandiflora **False pimpernel, savannah**
CA006p, CT007p, NE011p, NY005p

Liquidambar styraciflua **Sweetgum**
AR002p, CA003p, CA004s, DE001p, DE002p, FL001p, FL003p, FL005p, FL009p, FL010p, FL012p, FL013p, FL014p, FL015p, FL016p, GA002p, IN001p, KY001p, LA001p, LA005p, LA007p, LA008s, MA001s, MD002p, MI004p, MN002p, MO002p, MO005p, NC001p, NC002p, NC005p, NJ002p, NJ004p, NJ005p, NJ007s, NJ008p, NY003p, NY004s, NY005p, OH001b, OH003p, OH004p, OH008p, OK001p, OK004p, OK005p, OR001p, OR003p, OR007p, OR008p, OR009p, OR010p, OR011p, OR013p, OR014p, OR016p, PA003p, PA006p, PA008s, PA009p, PA010p, PA012p, SC002p, SD003p, TN001p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN010p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, VA002p, VA004p, VA005p

Liriodendron tulipifera **Tuliptree**
AL002p, AR002p, CA004s, CO005p, DE002p, FL001p, FL005p, FL009p, FL010p, FL012p, FL014p, FL015p, FL016p, IA001p, IA004p, IL003p, IL005p, IL006p, IN001p, KY001p, KY002p, LA001p, LA004p, LA005p, LA008s, MA001s, MD002p, MI003b, MI004p, MN006p, MN014p, MO002p, MO005p, NC001p, NC002p, NJ001p, NJ004p, NJ008p, NY003p, NY004s, OH001s, OH008p, OK001p, OK004p, OR005p, OR007p, OR008p, OR009p, OR010p, OR011p, OR014p, OR016p, PA003p, PA006p, PA009p, PA010p, PA012p, TN001p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN010p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, UT002p, VA002p, VA004p, VA005p

Litsea aestivalis **Pondspice**
FL014p, SC003p, VA005p

Lobelia anatina **Lobelia, Apache**
NJ007s

Lobelia cardinalis **Cardinalflower**

AZ004s, CA002p, CO003p, CO005p, CO006s, CT002p, CT003p, CT004p, CT009b, FL012p, FL014p, FL015p, IA003s, IL001s, IL004b, IN003p, KY002b, KY003p, LA002p, LA003p, MA002p, MA003p, MA004p, MD002p, MI001p, MI004p, MI005p, MN002p, MN003p, MN006p, MN009b, MN010p, NC003p, NC004p, NC006b, NE003s, NE011p, NJ001p, NJ005p, NJ007s, NJ008p, NY004s, NY005p, OH002p, OH005p, OH007p, OH008p, OK002s, OR014p, OR016p, PA004p, PA008s, PA009p, PA010p, PA012p, SC001p, SC002p, SC003p, SC004p, TN002p, TN003p, TN006p, TN010p, TN013p, TN015p, TN017p, TN019p, TN022p, TN023p, TN024p, TN025b, VA003p, VA004p, VA005p, WI001p, WI006p, WI008b, WI009b, WI010p, WI012p, WV001p

Lobelia dunnii **Lobelia, Dunn's**

CA002p

Lobelia elongata **Lobelia, elongated**

SC003p

Lobelia glandulosa **Lobelia, glade**

FL015p

Lobelia puberula **Lobelia, downy**

FL015p, NC005p, WV001p

Lobelia siphilitica **Lobelia, great blue**

CA006p, CO005p, CO006s, CT002p, CT003p, CT004p, FL012p, IA003s, IL001s, IL004b, IN003p, KY002b, KY003p, MA004p, MD002p, ME001s, MN002p, MN004b, MN009b, MN010p, MO003b, NC003p, NC004p, NC006p, NE003s, NE011p, NJ001p, NJ005p, NJ007s, NJ008p, NY002p, NY004s, NY005p, OH002p, OR004p, OR014p, OR016p, OH005p, PA008s, PA009p, PA010b, PA012p, SC002p, SC003p, TN003p, TN010p, TN013p, TN015p, TN017p, TN019p, TN022p, TN023p, TN025p, VA003p, VA004p, VA005p, WI001p, WI006p, WI008b, WI009b, WI010p, WI012b, WV001p

Lolium arundinaceum **Fescue, tall**

AR003s, AL004s, AL005s, AZ001s, CA014s, CA015s, CO006s, CO007s, CO009s, GA003s, GA005s, IA003s, IA004s, ID005s, ID006s, IL002s, KS001s, KS003s, MN015s, MO001s, MO006s, MO007s, MO008s, MT004s, MT008s, MT009s, MT013s, MT014s, NE002s, NE004s, NE005s, NE010s, NJ003s, NM001s, OK002s, PA008s, SD001s, SD002s, SD004s, TX009s, UT001s, UT004s, UT005s, UT006s, WA004s, WA005s, WY001s

Lonicera caerulea **Honeysuckle, sweetberry**

CT004p, MA004p

Ludwigia alternifolia **Seedbox, bushy**

IL004b, MN009s, PA009p, PA010b, PA012p

Ludwigia peploides **Primrose-willow, floating**

CA002p

Ludwigia repens **Primrose-willow, creeping**

FL016p, MD001p

Lupinus polyphyllus **Lupine, large-leaved**
CO001p, CO006s, CT003p, MA001s, ME001s, NE003s, NJ005p, NY004s, OH010p, OR011p, OR014p, PA002s, SC004p, UT001s, UT002p, WI005p, WI013p, WV001p

Lycium carolinianum **Desert-thorn, Carolina**
FL008p, FL015p, SC003p

Lycopus americanus **Water horehound, American**
MN009s

Lycopus europaeus **Gypsywort**
KY003p

Lycopus virginicus **Water horehound, Virginia**
PA010p

Lygodium japonicum **Fern, Japanese climbing**
WA001p

Lygodium palmatum **Fern, American climbing**
TN015p, TN023p

Lyonia ligustrina **Maleberry**
FL015p, MD002p, NJ005p, OR014p, PA008s

Lyonia lucida **Fetterbush**
FL001p, FL005p, FL010p, FL015p, FL016p, LA002p, OR014p, SC003p

Lysichiton americanus **Skunkcabbage, American**
CA017b, WI012b

Lysimachia ciliata **Loosestrife, fringed**
AR005p, CO005p, CT002p, CT004p, GA001p, IL004p, MN004b, MN009b, MN010p, NE011p, NY002p, OH002p, OH008p, OR004p, OR014p, VA003p

Lysimachia hybrida **Loosestrife, lowland yellow**
MN009b

Lysimachia nummularia **Jenny, creeping**
CA006p, CO001p, CO003p, CO005p, CT002p, CT004p, IL004p, LA002p, LA003p, MD001p, MI001p, MN002p, MN006p, MN010p, NC004p, NE011p, NY002p, OH002p, OH005p, OR004p, OR011p, OR012p, PA001p, PA010p, UT002p, VA003p, WI002p, WV001p

Lysimachia punctata **Loosestrife, large yellow**
CO005p, CT002p, CT003p, CT004p, IL004p, MN010p, NY002p, NY004s, NY005p, OH002p, OH005s, OH007p, OR011p, OR014p, VA003p, WI010p, WI013p, WV001p

Lysimachia quadriflora **Loosestrife, fourflower yellow**
IL004b, MN009b, PA008s

Lysimachia terrestris **Loosestrife, earth**
CT009s, PA004p

Lysimachia vulgaris **Loosestrife, garden yellow**
GA001p, WI005p

Lythrum alatum **Loosestrife, winged**
IL004b, MN009b

Macbridea caroliniana **Birds-in-a-nest, Carolina**
NC004p

Machaeranthera bigelovii* var. *bigelovii **Tansyaster, Bigelow's**
AZ004s, CO002p, UT001s

Magnolia virginiana **Magnolia, sweetbay**
AL001p, AR002p, AR005p, CA004s, DE001p, DE002p, FL001p, FL003p, FL004p, FL005p, FL007p, FL010p, FL012p, FL014p, FL015p, FL016p, IN001p, LA001p, LA002p, LA003p, LA007p, LA008s, MA004p, MD002p, MS002p, NC002p, NC005p, NJ001p, NJ004p, NJ005p, NJ008p, NY003p, NY004s, NY005p, OH001p, OH004p, OH008p, OR009p, OR014p, OR016p, PA001p, PA004p, PA009p, PA012p, SC001p, SC003p, TN002p, TN003p, TN004p, TN005p, TN007p, TN009p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN022p, TN024p, VA002p, VA004p, VA005p

Maianthemum stellatum **False lily of the valley, starry**
ID002s, MN009p, OR004p, OR014p

Maianthemum trifolium **False lily of the valley, three-leaf**
MN001p

Marsilea macropoda **Bigfoot waterclover (fern)**
TX004p

Marsilea quadrifolia **European waterclover (fern)**
NE011p

Matteuccia struthiopteris **Fern, ostrich**
AR002p, CA007p, CO001p, CO003p, CO005p, CT001p, CT002p, CT003p, CT004p, FL012p, FL016p, GA001p, IA004p, IL003p, IL004p, IN001p, MA003p, MA004p, MI001p, MI004p, MN002p, MN006p, MN009p, MN010p, NC005p, NE011p, NJ001p, NY002p, NY005p, OH005p, OH008p, OR004p, OR011p, OR014p, PA002p, PA010p, SC001p, SC002p, SD003p, TN013p, UT002p, VA003p, WA001p, WI002p, WI008p, WI010p, WI012p, WI013p

Mazus pumilus **Mazus, Japanese**
NC004p

Melanthium virginicum **Bunchflower, Virginia**
MN009b

Mentha aquatica **Mint, water**
MD001p, NC008p, NE011p, OH006p, PA002s, PA010p, WV001p

Mentha arvensis **Mint, wild**
CA002p, IN003p, MN009b, PA010p

Mentha pulegium **Pennyroyal**
CT002p, FL012p, IL004p, KY003p, ME001s, ME003s, NE003s, NE011p, OH005b, OH006p, PA002s, SC004s, WV001p

Mentha spicata **Spearmint**
CA004s, CT002p, CT003p, FL012p, IA004s, IL004p, ME001p, MN002p, NE011p, NY004s, OH005b, OH006p, PA002s, PA010p, SC004b, VA003p, WI002s, WI005p, WV001p

Mentha suaveolens **Mint apple**
KY003p, NE011p, NY005p, OH002p, OH005p, OH006p, SC001p, WV001p

Mentha X piperita **Peppermint**
CA004s, CT002p, CT003p, FL012p, IL004p, KY003p, ME001p, ME003p, MN002p, MN014s, NE011p, NJ007s, OH005b, OH006p, PA010p, SC001p, SC004b, TX007s, VA003p, WV001p

Menyanthes trifoliata **Buckbean**
CT004p, MN001p, OH008p

Mertensia bella **Bluebells, beautiful**
OR011p

Mertensia virginica **Bluebells, Virginia**
AR005p, CO001p, CT002p, CT003p, CT004p, IA004p, IL001s, IL004p, KY002p, KY003p, MI004p, MI005p, MN003p, MN006p, MN009p, MN010p, NC005p, NE011p, NY005p, OH005p, OH010p, OR004p, OR014p, PA008s, PA010p, TN003p, TN006p, TN010p, TN013p, TN015p, TN017p, TN019p, TN022p, TN023p, TN024p, TN025p, VA003p, WI002p, WI010p, WI011p

Mikania scandens **Hempweed, climbing**
KY002p, SC003p

Mimulus alatus **Monkeyflower, sharpwing**
KY002p

Mimulus cardinalis **Monkeyflower, scarlet**
CA002p, CA005p, CA007p, CA009p, CA013p, CA017b, CO005p, NJ005p, NJ007s, OR004p, OR011p, OR014p

Mimulus guttatus **Monkeyflower, seep**
CA002p, CA007p, CA009p, CA010s, CA013p, CA017b, CO003p, CO005p, MT001p, OR004p, OR014p

Mimulus lewisii **Monkeyflower, purple**
CA002p, CA007p, CO003p, CO004p, CO005p, CT002p, ID004s, MT001p, NJ007s, OR014p, UT002p

Mimulus primuloides **Monkeyflower, primrose**
OH005p

Mimulus ringens **Monkeyflower, Alleghany**
CT009b, IL001s, IL004b, IN003p, MA002p, MA003p, MN004b, MN009b, NC005p, NE003s, NE011p, NJ001p, NJ005p, NJ008p, OH008p, PA008s, PA009p, PA010b, PA012p, VA003p, VA004p, WI008s

Miscanthus sinensis **Silvergrass, Chinese**
AR002p, CA004s, CA006p, CA007p, CO001p, CO003p, CT001p, CT002p, CT004p, CT006p, DE002p, FL010p, FL012p, FL016p, GA001p, IA004p, IL004p, IN001p, LA001p, LA007p, MA004p, MD003p, MI001p, MI002p, MI004p, MI005p, MN002p, MN003p, MN006p, MN010p, NC002p, NC004p, NC008p, NE011p, NY002p, NY003p, NY004s, NY005p, OH001p, OH002p, OH003p, OH005p, OH007p, OH008p, OH009p, OH010p, OK001p, OR008p, OR011p, OR014p, PA002p, SC001p, SC002p, SC004b, SD003p, TN002p, TN004p, TN012p, UT002p, VA002p, VA003p, VA005p, WA002p, WI002p, WI003p, WI010p, WI013p, WV001p

Mitella pentandra **Miterwort, fivestamen**
NY005p

Monardella odoratissima **Monardella, mountain**
CA005p, CA007p, NJ007s

Montia parvifolia **Miner's-lettuce, little-leaf**
OR002p, OR004p

Morella (Myrica) californica **Bayberry, Pacific**
CA002p, CA005p, CA006p, CA007p, CA008s, CA009p, CA013p, CA017b, NY004s, OR008p, OR009p, OR011p

Morella (Myrica) caroliniensis **Bayberry, southern**
FL014p, FL015p, NY004s, OR014p, SC003p

Morella (Myrica) cerifera **Myrtle, wax**
AR002p, CA004s, FL001p, FL003p, FL004p, FL005p, FL006p, FL008p, FL009p, FL010p, FL012p, FL014p, FL016p, GA002p, ID002s, LA001p, LA002p, LA003p, LA007p, LA008s, MA001s, MD002p, MS002p, NC001p, NC002p, NJ005p, NJ008p, NJ009p, NY004s, OK001p, OR014p, OR014p, PA004p, PA009p, PA010p, SC001p, SC003p, VA002p, VA004p, VA005p

Morella (Myrica) gale **Sweetgale**
NY004s, OR014p

Morella (Myrica) inodora **Bayberry, odorless**
FL015p, NY004s, SC003p

Muhlenbergia capillaris **Muhly, hairawn**
CA006p, CA007p, FL003p, FL006p, FL008p, FL009p, FL012p, FL015p, FL016p, GA001p, NC004p, NC008p, OK001p, SC001p, SC003p, TX004p

Muhlenbergia glomerata **Muhly, spiked**
IL004b, MN004b

Muhlenbergia lindheimeri **Muhly, Lindheimer's**
CA006p, CA007p, NC008p, SC001p, TX004p

Muhlenbergia mexicana **Muhly, Mexican**
IL004b, MN009b

Muhlenbergia rigens **Deergrass**
CA002p, CA005p, CA006p, CA007p, CA009p, CA010s, CA013p, OR014p, TX004p

Musa acuminata **Banana, edible**
FL005p, FL012p, LA007p

Myosotis asiatica **Forget-me-not, Asian**
CO001p, CO005p, CT002p, MN002p, MN003p, NY005p, OH002p, OH005b, SC002p, SC004p, VA003p, WI010p

Myosotis scorpioides **Forget-me-not, true**
CT002p, NC003p, NE011p, NY005p, OH007p, PA010p, SC001p, VA003p, WI010p

Myosotis sylvatica **Forget-me-not, woodland**
CO006s, CT003p, MI002p, MI005p, MN010p, MT015p, NE011p, OH010p, OR008p, SC004b, SD003s, WI013p

Myriophyllum aquaticum **Watermilfoil, parrot feather**
CA017p, CT004p, MD001p, NC003p, NC008p

Myriophyllum humile **Watermilfoil, low**
PA004p

Myrsine floridana **Guianese collicwood**
FL003p, FL004p, FL005p, FL008p, FL016p

Napaea dioica **Glademallow**
IL004b, MN009b, NE003s, WI008b

Nelumbo lutea **Lotus, American**
CT004p, IL002s, MD001p, NC003p, NE003s, PA010p, VA005p, WI001s, WI007s, WI012b

Nelumbo nucifera **Lotus, sacred**
MD001p, NC008p

Nemopanthus mucronatus **Catberry**
CT005p, MA003p, PA008s

Nepeta cataria **Catnip**
CA004s, CT002p, CT003p, FL012p, IA004s, ID004s, IL001s, IL004p, KY003p, ME001s, ME003s, MN001p, MN002p, NE003s, NJ007s, OH005b, OH006p, PA002s, SC004s, SD003s, TX007s, VA003p, WI002s, WV001p

Nephrolepis biserrata **Swordfern, giant**
FL005p, FL008p, FL012p, OH005p

Nuphar lutea **Pond-lily, yellow**
CO005p, FL016p, FL017p, IN003p, MA003p, MD002p, NC003p, NJ008p, OR009p, PA004p, PA010p, PA012p, VA004p, VA005p, WI007p, WI012p

Nymphaea capensis **Waterlily, Cape Blue**
CT004p

Nymphaea mexicana **Waterlily, banana**
NC003p

Nymphaea odorata **Waterlily, American white**
CT009p, FL016p, FL017p, GA001p, IN003p, KY002p, MA003p, MA004p, MD001p, NC003p, NC008p, NJ005p, OR009p, PA004p, PA010p, PA012p, VA005p, WI001p, WI007p, WI012p

Nymphaea tetragona **Waterlily, pygmy**
NC003p

Nymphoides peltata **Floatingheart, yellow**
MD001p

Nyssa aquatica **Water-tupelo**
AL007p, FL001p, FL010p, FL014p, FL016p, GA002p, LA005p, LA008s, MA001s, MD002p, MS001p, NC001p, NY004s, OR014p, PA004p, SC003p, TN007p, TN017p, VA005p

Nyssa biflora **Tupelo, swamp**
FL001p, FL009p, FL010p, FL014p, FL015p, FL016p, OR014p, SC003p

Nyssa ogeche **Tupelo, Ogeechee**
FL014p, FL015p, LA008s, NY004s, OR014p

Oenanthe sarmentosa **Water parsley**
CA017b, OR009p

Oenothera elata **Evening-primrose, Hooker's**
AR005p, CA013p

Oligoneuron ohioense **Goldenrod, Ohio**
IL004b, IN003p, MN009s, WI009b

Oligoneuron riddellii **Goldenrod, Riddell's**
AR005p, IL004b, IN003p, MN009b, NC005p, PA008s, WI008b

Onoclea sensibilis **Fern, sensitive**
CT002p, CT009p, FL005p, GA001p, IL004p, KY002p, MA003p, MA004p, MI004p, MN001p, MN009b, NJ001p, NJ008p, NY005p, OH008p, OR004p, OR014p, PA004p, PA008s, PA010p, PA012p, SC001p, TN002p, TN003p, TN007p, TN010p, TN013p, TN015p, TN017p, TN019p, TN022p, TN023p, TN024p, TN025p, VA004p, WA001p, WI006p

Oplopanax horridus **Devilsclub**
OR009p

Oreostemma alpigenum* var. *alpigenum **Aster, tundra**
CA007p, NY002p

Orontium aquaticum **Goldenclub**
CT009p, KY002p, MD001p, NC003p, NC004p, NC005p, NC008p, NJ001p, PA004p, PA010p, VA005p

Oryza sativa **Rice (cultivated)**
AR003s, GA002s, GA003s, IL002s, MO011s

Osmunda cinnamomea **Fern, cinnamon**
AR002p, CO001p, CO005p, CT001p, CT002p, CT004p, CT009p, FL005p, FL010p, FL012p, FL016p, GA001p, IA004p, IL003p, IL004p, IL006p, IN001p, KY002p, MA003p, MA004p, MI001p, MN010p, NC004p, NE011p, NJ001p, NJ008p, NY002p, NY005p, OH003p, OH005p, OH008p, OR011p, OR014p, PA004p, PA010p, PA012p, SC001p, SC002p, SC003p, SD003p, TN002p, TN003p, TN006p, TN007p, TN010p, TN013p, TN015p, TN017p, TN019p, TN022p, TN023p, TN024p, VA003p, VA004p, VA005p, WI002p, WI010p, WI012p, WI013p

Osmunda regalis **Fern, royal**
AR002p, CT002p, CT004p, FL005p, FL010p, FL012p, FL016p, GA001p, IL004p, LA003p, MA003p, MA004p, MI001p, MN010p, NC005p, NE011p, NJ001p, NJ008p, NY002p, NY005p, OH005p, OH008p, OR004p, OR011p, OR014p, PA004p, PA010p, PA012p, SC001p, SC003p, TN002p, TN003p, TN006p, TN007p, TN010p, TN013p, TN017p, TN019p, TN022p, TN023p, TN024p, VA003p, VA004p, VA005p, WA001p, WI010p, WI012p

Oxyria digyna **Mountainsorrel, alpine**
MT001p

Panicum anceps **Panicgrass, beaked**
NJ005p

Panicum dichotomiflorum **Panicgrass, fall**
IL002s, PA008s

Panicum hemitomon **Maidencane**
FL005p, FL010p, FL016p, FL017p, LA006p, MS003p, SC005p

Panicum obtusum **Vine mesquite**
NM001s, TX001s, UT001s

Panicum repens **Grass, torpedo**
MS003p

Panicum virgatum **Switchgrass**

AR002p, AR004p, AR005p, CA004s, CA006p, CA007p, CO003p, CO004p, CO005p, CO006s, CO007s, CO009s, CT002p, CT003p, CT004p, CT006p, DE002p, FL016p, GA001p, GA002s, GA003s, IA003s, IA005s, ID002s, ID006s, IL002s, IL004b, IN001p, IN003p, KS001s, KS003s, KS005s, KY002b, MA002p, MA003p, MA004p, MD002p, ME001s, MI001p, MI004p, MN002p, MN003p, MN004b, MN006p, MN004b, MN006p, MN007s, MN009b, MN010p, MN011s, MN013s, MO001s, MO006s, MO007s, MO009s, NC004p, NC008p, ND001s, ND002s, NE002s, NE003s, NE004s, NE005s, NE006s, NE008s, NE009s, NE011p, NJ001p, NJ008p, NJ009p, NM001s, NY002p, NY004s, NY005p, NY007s, OH002p, OH003p, OH007p, OH008p, OK002s, OR008p, OR011p, OR014p, PA001p, PA008s, PA009p, PA010b, PA012p, SC001p, SC002p, SD001s, SD002s, TN013p, TN025p, TX001s, TX002s, TX004p, TX005s, TX009s, TX010s, TX011s, TX012s, TX016s, TX017s, UT001s, UT002p, UT004s, UT005s, UT006s, VA001p, VA003p, VA004p, VA005p, WA005s, WI001s, WI007s, WI008b, WI009b, WI010p, WI012p, WV001p, WY001s

Parkinsonia aculeata **Jerusalem thorn**

CA004s, CA005p, FL005p, FL016p, MA001s, NY004s

Parnassia asarifolia **Grass of Parnassus, kidneyleaf**

TN024p

Parnassia glauca **Grass of Parnassus, waxy**

MN009s, TN017p

Parnassia palustris **Grass of Parnassus, northern**

MN001p

Paspalum vaginatum **Paspalum, seashore**

FL005p, FL017p, LA006p

Pedicularis groenlandica **Lousewort, elephanthead**

ID002s, ID004s, MT004s, WY001s

Pedicularis lanceolata **Lousewort, swamp**

MN009s, WI008s

Peltandra virginica **Arum, green arrow**

CT004p, CT009p, FL016p, MA003p, MD002p, NC003p, NC008p, NJ005p, NJ006p, NJ008p, OH008p, PA004p, PA008s, PA009p, PA010b, PA012p, VA001p, VA004p, VA005p, WI001s, WI007p, WI012p

Penstemon digitalis **Beardtongue, foxglove**

AR002p, AR005p, CA007p, CO001p, CO003p, CO005p, CO006s, CT001p, CT002p, CT003p, CT004p, FL012p, GA001p, IA002p, IL004p, KY002b, LA002p, ME001s, ME003p, MI001p, MI002p, MN002p, MN003p, MN006p, MN009b, MN010p, MO003b, NC004p, NC005p, NE003s, NE011p, NJ001p, NJ007s, NY002p, NY005p, OH002p, OH005p, OH007p, OH008p, OK001p, OR004p, OR014p, PA008s, SC001p, SC003p, SC004p, TN013p, UT002p, VA002p, VA003p, WI002p, WI008b, WI009b, WI010p, WI013p

Penstemon tenuis **Beardtongue, sharpsepal**

AR005p

Pentaphylloides floribunda **Cinquefoil, shrubby**
CA009p, CO001p, CO003p, CO004p, CO005p, CT001p, CT002p, CT004p, CT006p, GA001p, IA002p, IA004p, IN001p, MI002p, MI004p, MN002p, MN003p, MN004b, MN006p, MN009p, MN014p, MN020p, MT001p, MT002p, MT005p, MT006p, MT007p, MT015p, NJ004p, NY003p, NY005p, NY006p, OH001p, OH002p, OH003p, OH008p, OH009p, OR012p, OR014p, OK001p, OK004p, OK005p, OR005p, OR008p, OR009p, OR011p, OR014p, PA001p, PA005p, SC002p, SD003p, TN003p, UT002p, VA005p, WI002s, WI003p, WI004p

Penthorum sedoides **Ditch stonecrop**
MN009s

Persea borbonia **Redbay**
FL003p, FL004p, FL005p, FL007p, FL008p, FL009p, FL015p, FL016p, LA002p, LA008s, NC005p, NC007p, NY004s, VA005p

Petasites frigidus **Coltsfoot, Arctic sweet**
OH014p

Petasites frigidus var. palmatus **Coltsfoot, Arctic sweet**
NY005p

Phalaris arundinacea **Reed canarygrass**
CA004s, CA006p, CA007p, CO001p, CO003p, CO005p, CO007s, CO009s, CT001p, CT002p, CT003p, CT004p, FL012p, FL016p, GA001p, GA002s, GA003s, IA003s, ID006s, IL002s, IL004p, IN001p, KS001s, LA007p, MA002p, MA004p, MI001p, MI002p, MI004p, MN002p, MN006p, MN007s, MN016s, MO001s, MO006s, MO007s, MO011s, MT003s, MT004s, NC008p, NE002s, NE005s, NE006s, NE007s, NE011p, NJ003s, NY002p, NY005p, OH002p, OH003p, OH005p, OH007p, OH008p, OR011p, OR014p, PA001p, PA008s, PA010b, PA012p, SC001p, SD001s, VA004p, VA005p, WA002p, WI001s, WI007s, WY001s, UT001s, UT002p,UT004s, UT005s, UT006s, VA003p, WY002s

Phleum alpinum **Timothy, alpine**
MT004s

Phlox carolina **Phlox, thickleaf**
AR002p, CO005p, CT002p, CT004p, GA001p, IL004p, KY002p, MA004p, MI001p, MN010p, NC004p, NE011p, OH002p, OR014p, TN025p, VA003p, WI002p, WI013p

Phlox glaberrima **Phlox, smooth**
CT002p, NC004p, NC006p, NY005p, OH002p, OH005p, SC003p, TN013p, VA003p

Phlox maculata **Wild sweetwilliam**
AR002p, CT002p, FL012p, GA001p, KY002p, MA004p, MI002p, MN002p, MN006p, MN009b, MN010p, NC004p, NC005p, NE011p, NY002p, NY005p, OH002p, OH005p, OH007p, OR014p, TN013p, TN024p, VA003p, WI010p

Photinia arbutifolia (Aronia arbutifolia) **Chokeberry, red**

AR002p, AR005p, CO001p, CO003p, CO005p, CT004p, CT005p, DE002p, FL015p, IA002p, IA004p, IN001p, MA003p, MD002p, MI004p, MN002p, MN006p, MT002p, MT005p, MT007p, NJ001p, NJ004p, NJ005p, NJ008p, NY003p, NY004s, NY006p, OH003p, OH008p, OK001p, OR005p, OR009p, OR014p, PA001p, PA004p, PA006p, PA008s, PA009p, PA010p, PA012p, SC003p, TN003p, TN007p, TN010p, TN013p, TN017p, TN018p, TN022p, UT002p, VA002p, VA004p, VA005p, WI002s, WI003p

Photinia melanocarpa (Aronia melanocarpa) **Chokeberry, black**

AR002p, CO001p, CO003p, CO005p, CT005p, DE002p, IA002p, ID003p, IN001p, KY002p, MA001s, MA003p, MD002p, MN002p, MN003p, MN006p, MN009p, MT007p, NC005p, NJ001p, NJ002p, NJ004p, NJ005p, NJ008p, NY003p, NY004s, NY005p, NY006p, OH003p, OH008p, OR009p, OR011p, OR014p, PA004p, PA005p, PA008s, PA009p, PA010p, PA012p, TN003p, TN010p, TN017p, TN018p, TN022p, TN024p, UT002p, VA004p, VA005p, WI002s, WI004p, WI006p

Photinia floribunda (Aronia x prunifolia) **Chokeberry, purple**

MA001s, MD002p, NJ005p, NY004s

Phragmites australis **Reed, common**

CO004p, IA005s, LA006p, MA002p, MA004p, NC008p, NE011p, OH008p, OK003p, PA004p, PA008s, PA010p, TX004p, TX006p, WI001p, WI007p, WI012p

Physocarpus capitatus **Ninebark, Pacific**

CA002p, CA009p, CA013p, CA017b, MT001p, OR009p, OR014p

Physocarpus opulifolius **Ninebark, common**

AR005p, CO001p, CO003p, CO005p, CT004p, IA002p, IN001p, MA001s, MN002p, MN003p, MN006p, MN009b, MN014p, MT006p, MT007p, MT015p, NY004s, NY006p, OK004p, OR005p, OR014p, PA004p, PA008s, PA009p, PA010p,TN002p, TN003p, TN008p, UT002p, WI002s, WI003p, WI004p, WI012p

Physostegia angustifolia **Dragonhead, narrowleaf false**

TX004p

Physostegia purpurea **Dragonhead, eastern false**

FL015p

Physostegia virginiana **Obedient plant**

AR002p, CO001p, CO003p, CO005p, CT002p, CT003p, GA001p, IA004p, IL004b, IN003p, KY002b, LA002p, LA003p, MA004p, MI001p, MI002p, MI004p, MI005p, MN002p, MN003p, MN006p, MN009b, MN010p, MO003b, NE011p, NJ001p, NJ005p, NJ007s, NY002p, NY005p, OH002p, OH003p, OH005b, OH007p, OH008p, OH010p, OR011p, OR014p, PA008s, SC001p, SC002p, SC003p, SC004b, SD003p, TN013p, TN025p, UT002p, VA003p, WI002p, WI005p, WI008b, WI009p, WI010p, WI013p

Picea mariana **Spruce, black**

CA003p, CT001p, MA001s, MA003p, ME002p, MI003p, MN002p, MN006p, MN017p, NY004s, NY005p, OH001p, OH008p, OR006p, OR011p, OR014p, PA004p, PA010p, PA011p

Pieris phillyreifolia **Fetterbush, climbing**
FL015p, SC003p

Pinckneya bracteata **Fevertree**
FL014p, FL015p, NC004p, NC007p, NY004s, OR014p, SC003p, VA005p

Pinguicula caerulea **Butterwort, blueflower**
NY001p

Pinguicula lutea **Butterwort, yellow**
NY001p

Pinus contorta var. contorta **Pine, lodgepole**
CA017b, MA001s, MT007p, NY004s

Pinus elliottii **Pine, slash**
AR002p, CA004s, DE002p, FL001p, FL005p, FL007p, FL014p, FL016p, FL017p, GA002p, LA001p, LA003p, LA005p, LA008s, MA001s, NY004s, OK001p, OK004p, OK005p, OR014p, TX014p

Pinus glabra **Pine, spruce**
AR002p, CA004s, FL005p, FL010p, FL014p, FL015p, FL016p, LA005p, LA007p, LA008s, MA001s, NY004s, OR014p

Pinus serotina **Pine, pond**
FL001p, FL005p, FL009p, FL015p, FL016p, MA001s, MD002p, NC001p, NY004s, OR014p, SC003p

Pistia stratiotes **Water lettuce**
CT004p, MD001p, NC003p

Planera aquatica **Planertree**
FL009p, FL015p, SC003p

Plantago major **Plantain, common**
NJ007s

Plantago maritima **Goose tongue**
CA017b

Plantago subnuda **Plantain, tall coastal**
CA009p

Platanthera blephariglottis **Orchid, white fringed**
NC005p

Platanthera ciliaris **Orchid, yellow fringed**
FL015p, NC005p

Platanus occidentalis ***Sycamore, American***
AL002p, AL007p, AR002p, CA004s, FL001p, FL005p, FL009p, FL010p, FL012p, FL014p, FL015p, FL016p, LA001p, LA003p, LA005p, LA007p, LA008s, MA001s, MD002p, MO002p, MT007p, NC001p, NC002p, NJ004p, NJ005p, NJ008p, NY003p, NY004s, OK001p, OK004p, OK007p, OR014p, PA008s, PA009p, PA010p, PA012p, TN001p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN010p, TN011p, TN012p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, TX008p, TX013p, TX014p, TX015p, VA003p, VA004p, VA005p

Platanus racemosa **Sycamore, California**
CA002p, CA004s, CA005p, CA006p, CA007p, CA013p, CA017b

Platanus wrightii **Sycamore, Arizona**
AZ002p, CA004s

Pluchea odorata **Sweetscent**
CA010s

Pluchea odorata* var. *odorata **Sweetscent**
CA010s

Pluchea sericea **Arrowweed**
CA002p

Poa annua **Bluegrass, annual**
IL002s, PA008s

Poa palustris **Bluegrass, fowl**
CO005p, MA002p, MA003p, MT004s, NJ003s, PA008s, PA010p, UT001s

Poa pratensis **Bluegrass, Kentucky**
AR003s, AZ001s, CA014s, CA015s, CO007s, IA004s, ID004s, ID005s, ID006s, IL002s, KS001s, NE002s, NJ003s, NM001s, MT003s, MT004s, OK002s, PA008s, SD001s, SD002s, TX010s, UT001s, UT004s, UT005s, UT006s, WY001s

Poa trivialis **Bluegrass, rough**
CA014s, IL002s, PA008s, UT001s

Pogonia ophioglossoides **Pogonia, rose (snakemouth orchid)**
FL015p

Polemonium pauciflorum **Jacob's-ladder, fewflower**
NJ007s

Polemonium vanbruntiae **Jacob's-ladder, sticky**
NJ001p, TN003p, TN015p, TN023p

Polygonum amphibium **Water knotweed**
CA017b, MO011p, MT001p, OR009p, PA010p, WI001p, WI007p, WI012p

Polygonum arifolium **Tearthumb, halberdleaf**
PA004p, PA008s

Polygonum bistorta **Bistort, meadow**
CT002p, KY003p, MN010p, OH008p, OR004p, OR014p, SC001p, UT002p

Polygonum hydropiperoides **Smartweed, swamp**
CA017b

Polygonum lapathifolium **Curlytop knotweed**
IL002s, PA010p, WI001s, WI008s

Polygonum orientale **Kiss me over the garden gate**
NE003s, NJ007s

Polygonum pensylvanicum **Smartweed, Pennsylvania**
GA003s, IL001s, IL002s, MO001s, NJ008p, OK003p, PA004p, PA008s, PA010b, VA004p, WI001s, WI008s, WI012s

Polygonum persicaria **Ladysthumb, spotted**
CA017b, IL002s, PA008s, PA010p

Polygonum punctatum **Smartweed, dotted**
OR009p, PA010p

Polygonum sagittatum **Tearthumb, arrowleaf**
MN009s

Polypogon monspeliensis **Grass, rabbitsfoot**
CO004p, PA010p

Pontederia cordata **Pickerelweed**
CT004p, CT009p, FL010p, FL016p, FL017p, IL002s, IN003p, KY002p, LA002p, LA003p, MA002p, MA003p, MA004p, MD001p, MD002p, NC003p, NC005p, NC008p, NJ006p, NJ008p, OH008p, PA004p, PA008s, PA009p, PA010p, PA012p, SC001p, VA001p, VA004p, VA005p, WI001p, WI007p, WI012b

Populus angustifolia **Cottonwood, narrowleaf**
CA002p, CO001p, CO003p, CO004p, CO005p, ID003p, MN002p, MT001p, MT006p, OR014p, PA010p, UT002p, UT003p

Populus balsamifera **Poplar, balsam**
CA017p, MN002p, OR014p

Populus deltoides **Cottonwood, eastern**
AR002p, CO001p, CO004p, CO005p, CO008p, IA001p, IA004p, LA005p, MA003p, MN002p, MN003p, MS001p, MT001p, MT007p, OK004p, OK005p, PA010p, PA012p, TX008p, TX013p, TX014p, UT002p, UT003p, VA005p, WI002s, WI003p

Populus fremontii **Cottonwood, Fremont**
CA002p, CA005p, CA006p, CA007p, CA017p, MA001s, MT001p, NV001p, OR014p, UT003p

Populus X acuminata **Cottonwood, lanceleaf**
CO003p, CO004p, CO005p, ID007p, MN002p, MT005p, OK004p, OR005p, PA010p

Potamogeton amplifolius **Pondweed, largeleaf**
PA004p, WI012p

Potamogeton nodosus **Pondweed, longleaf**
MA003p, PA010p, WI001p

Potamogeton richardsonii **Pondweed, Richardson's**
WI001p, WI012p

Potamogeton robbinsii **Pondweed, Robbins'**
PA004p

Potentilla glandulosa **Cinquefoil, gland**
CA007p

Potentilla gracilis **Cinquefoil, slender**
CA002p, CA009p

Potentilla thurberi **Cinquefoil, scarlet**
CT002p, NJ007s, SC004s

Prenanthes racemosa **Rattlesnakeroot, purple**
MN009b

Primula parryi **Primrose, Parry's**
CT002p, OR014p

Prosopis pubescens **Mesquite, screwbean**
CA004s

Prunella vulgaris **Heal-all (common selfheal)**
KY003p, LA002p, ME001s, NE011p, WV001p

Psychotria nervosa **Seminole balsamo**
FL002p, FL003p, FL004p, FL005p, FL008p, FL015p, FL017p

Psychotria tenuifolia **Coffee, shortleaf wild**
FL004p, FL015p

Puccinellia distans **Weeping alkaligrass**
CO004p, CO007s, ID002s, MN018s, MT010s, MT013s, NJ003s, PA008s, UT001s, UT004s, WY001s

Puccinellia lemmonii **Lemmon's alkaligrass**
ID006s

Puccinellia nuttalliana **Nuttall's alkaligrass**
CO004p, MT001p, MT004s, PA010p, UT001s, UT004s, WY001s

Pycnanthemum muticum **Mountainmint, clustered**
MA004p, OH006p, PA008s

Pycnanthemum tenuifolium **Mountainmint, narowleaf**
AR005p, IA003s, KY002b, MN009s, MO003b, NJ001p, PA008s, SC003p

Pycnanthemum virginianum **Mountainmint, Virginia**
AR005p, IL004b, IN003p, MI008b, MN004b, MN009b, MN011s, NE011p, OH006p, WI008s, WI009s

Pyrola asarifolia **Wintergreen, pink**
ID002s

Quercus bicolor **Oak, swamp white**
CA004s, CO003p, CO005p, DE001p, DE002p, GA002p, IA001b, ID007p, IN001p, IN002p, KY001p, MA001s, MA003p, MD002p, ME002p, MI003p, MN002p, MN003p, MN005p, MN006p, MN009p, MN012p, MN017p, MO002p, MO005p, MT007p, NJ002p, NJ004p, NJ005p, NJ008p, NY004s, OH004p, OH008p, OK001p, OR001p, OR003p, OR005p, OR009p, OR014p, OR016p, PA003p, PA004p, PA005p, PA006p, PA009p, PA010p, PA012p, TN003p, TN004p, TN007p, TN011p, TN017p, TN020p, TN022p, UT002p, VA004p, VA005p, WI002s, WI003p

Quercus laurifolia **Oak, laurel**
CA004s, FL001p, FL003p, FL005p, FL007p, FL010p, FL014p, FL015p, FL016p, LA008s, NC002p, NY004s, OR014p, SC003p

Quercus lyrata **Oak, overcup**
AL007p, AR002p, FL001p, FL010p, FL014p, FL015p, GA002p, LA004p, LA005p, LA008s, MA001s, MO002p, MO005p, MS001p, MS005p, NC001p, NY004s, OR014p, PA012p, SC003p, TN003p, TN007p, TN017p, TN020p, VA005p

Quercus michauxii **Oak, swamp chestnut**
AL007p, AR002p, FL001p, FL009p, FL014p, GA002p, LA002p, LA004p, LA005p, LA008s, MI003p, MO001s, MO002p, MO005p, MS001p, MT007p, NC001p, NY004s, OR014p, PA012p, SC003p, TN003p, TN007p, TN017p, VA005p

Quercus nigra **Oak, water**
AL002p, AL006p, AL007p, AR001p, AR002p, AR005p, FL001p, FL005p, FL009p, FL010p, FL014p, FL015p, FL016p, GA002p, LA001p, LA004p, LA005p, LA008s, MO002p, MS001p, MS004p, NC001p, NY004s, OK001p, OK005p, OR014p, PA010p, TN002p, TN003p, TN007p, TN010p, TN011p, TN017p, TN018p, TN019p, TN021p, TN022p, TN024p, TX013p, TX014p, VA005p

Quercus pagoda **Oak, cherrybark**
AL007p, AR001p, AR002p, FL001p, FL014p, FL015p, GA002p, LA001p, LA002p, LA004p, LA005p, LA008s, MO002p, MO005p, MS001p, MS004p, NC001p, NC002p, NY004s, OR014p, PA012p, SC006p, TN003p, TN007p, TN008p, TN017p, TN020p, TN024p, TX014p

Quercus palustris — **Oak, pin**

AL002p, AL006p, AR002p, AR005p, CA004s, CO003p, CT001p, DE001p, DE002p, FL001p, GA002p, IA001b, ID001p, IN001p, KY001p, LA001p, LA008s, MA001s, MA003p, MD002p, MI003p, MI004p, MN002p, MN003p, MN005p, MN006p, MN012p, MO002p, MO005p, MS001p, MT007p, NC002p, NJ002p, NJ004p, NJ005p, NJ008p, NY003p, NY004s, NY006p, OH003p, OH004p, OH008p, OK001p, OK004p, OK005p, OR001p, OR003p, OR005p, OR007p, OR008p, OR009p, OR014p, PA003p, PA004p, PA005p, PA006p, PA007p, PA008s, PA009p, PA010p, PA011p, PA012p, SD003p, TN001p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN010p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, TX014p, UT002p, VA002p, VA004p, VA005p, WI003p

Quercus phellos — **Oak, willow**

AL007p, AR001p, AR002p, CA004s, DE002p, FL001p, FL014p, GA002p, IA001p, LA001p, LA003p, LA004p, LA005p, LA008s, MA001s, MD002p, MO002p, MS001p, MS004p, MS005p, NC001p, NC002p, NJ008p, NY003p, NY004s, OK001p, OR009p, OR010p, OR011p, OR014p, PA009p, SC003p, TN001p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN010p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, VA002p, VA004p, VA005p

Quercus shumardii — **Oak, Shumard**

AL001p, AL006p, AR001p, AR002p, CA004s, CO003p, DE002p, FL001p, FL003p, FL009p, FL012p, FL014p, FL015p, FL016p, GA002p, LA001p, LA003p, LA004p, LA005p, LA007p, LA008s, MA001s, MI003p, MN002p, MO002p, MO005p, MS001p, MS004p, MT007p, NC002p, NJ004p, NY004s, OH004p, OH008p, OK001p, OK004p, OK005p, OK007p, OR001p, OR009p, OR014p, SC003p, TN001p, TN002p, TN003p, TN004p, TN006p, TN007p, TN008p, TN016p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, TX003p, TX013p, TX014p, UT002p

Quercus texana (nuttallii) — **Oak, Texas red (Nuttall oak)**

AR001p, AL007p, AR002p, DE002p, FL001p, FL010p, FL014p, GA002p, LA002p, LA004p, LA005p, LA007p, LA008s, MO002p, MO005p, MS001p, MS004p, MS005p, NY004s, OK001p, OK005p, OR014p, TN003p, TN007p, TN008p, TN017p, TN018p, TN022p, TN024p, TX004p, TX014p

Ranunculus abortivus — **Buttercup, littleleaf**

IL002s

Ranunculus acris — **Buttercup, tall**

CT002p, IL001s, MI001p, MN010p

Ranunculus flabellaris — **Buttercup, yellow water**

PA010p, WI001p, WI012p

Ranunculus flammula — **Greater creeping spearwort**

OH008p

Ranunculus hispidus — **Buttercup, bristly**

MN009b, MO003b

Ranunculus hispidus var. nitidus — **Buttercup, bristly**

PA004p

Ranunculus longirostris **Buttercup, longbeak**
WI012p

Ranunculus occidentalis **Buttercup, western**
CA017b

Ranunculus pensylvanicus **Buttercup, Pennsylvania**
MN009b

Ranunculus repens **Buttercup, creeping**
CA017b, CO003p, CO005p, MI001p, MN010p, NC005p, NE011p, OH005p, OR009p, OR014p, PA010p, SC002p

Rhamnus alnifolia **Buckthorn, alderleaf**
ID002s, OR014p

Rhapidophyllum hystrix **Palm, needle**
CA004s, FL003p, FL004p, FL014p, FL012p, FL015p, SC003p, VA005p

Rhexia alifanus **Meadowbeauty, savannah**
LA002p

Rhexia lutea **Meadowbeauty, yellow**
NC005p

Rhexia mariana **Meadowbeauty, Maryland**
NC005p

Rhexia virginica **Handsome Harry**
NC004p, NJ001p

Rhizophora mangle **Mangrove, American**
FL002p, FL005p, FL006p, FL008p, FL016p

Rhododendron arborescens **Azalea, smooth**
FL015p, NC004p, NC005p, NY003p, NY005p, OH001b, OH002p, OH008p, OR002p, OR011p, PA012p, SC003p, TN005p, TN013p

Rhododendron canadense **Rhodora**
NC005p, NY004s, NY005p, OH001p, OH008p, OR011p

Rhododendron canescens **Azalea, mountain**
AR002p, FL014p, FL015p, NC004p, NC005p, NY005p, OR002p, OR014p, SC003p, VA005p

Rhododendron chapmanii **Rhododendron, Chapman's**
FL015p, LA002p, SC003p

Rhododendron oblongifolium **Azalea, Texas**
FL015p, OH001p, OR011p

Rhododendron viscosum **Azalea, swamp**
CT004p, CT005p, FL015p, LA002p, MA001s, MA003p, MA004p, MD002p, MN002p, NC005p, NE003s, NJ001p, NJ008p, NY003p, NY004s, NY005p, OH001b, OH002p, OH008p, OH009p, OR011p, OR014p, PA001p, PA012p, SC003p, TN013p, VA004p, VA005p

Rhynchospora capitellata **Beakrush, brownish**
CT009s

Rhynchospora colorata **Starrush whitetop**
SC001p

Rhynchospora latifolia **Sandsamp whitetop**
NC004p

Ribes aureum **Currant, golden**
CA002p, CA006p, CA007p, CA009p, CA017b, CO001p, CO003p, CO004p, CO005p, ID001p, ID002s, MN002p, MT001p, MT002p, MT005p, MT006p, MT007p, NV001p, OH008p, OR009p, OR011p, OR010p, OR014p, UT001s, UT002p, UT004s, UT006p, WA003p, WY001s

Ribes divaricatum **Gooseberry, spreading**
CA002p, CA013p

Ribes hirtellum **Gooseberry, hairystem**
AR002p, CO001p, CO003p, CT008p, GA001p, IA004p, IL003p, IL004p, IL005p, MI001p, MN002p, MN006p, MN014p, MO004p, MT002p, MT005p, MT006p, NY003p, NY006p, OR001p, SD003p, TN006p, WI002p, WI003p

Ribes inerme **Gooseberry, whitestem**
CO003p, NY004s

Ribes lacustre **Currant, prickly**
OR009p, OR014p

Rorippa nasturtium-aquaticum **Watercress**
CA017p, FL012p, IL002s, MN002p, NE003s, OH005b, PA010p, WI001s

Rosa eglanteria **Rose, sweetbriar**
MA001s, NE003s, NY004s, OR014p

Rosa nitida **Rose, shining**
IL001s, OR014p

Rosa palustris **Rose, swamp**
FL015p, IL004p, LA002p, MA003p, MD002p, MN009b, NH001p, NJ001p, NJ008p, NY004s, OR014p, PA004p, PA008s, PA009p, PA012p, VA004p, VA005p

Rubus hispidus **Dewberry, bristly**
NJ005p

Rudbeckia californica **Coneflower, California**
NJ007s, OR014p

Rudbeckia fulgida **Coneflower, orange**

AR005p, CO001p, CO003p, CO005p, CT001p, CT002p, CT003p, CT004p, DE002p, FL012p, FL014p, GA001p, IA002p, IL003p, IL004p, IL005p, IL006p, IN003p, KY002p, KY003p, MD003p, ME003p, MI001p, MI002p, MI004p, MN002p, MN003p, MN006p, MN009b, MN010p, MN020p, NC004p, NC005p, NC006p, NE011p, NJ001p, NJ007s, NY002p, NY004s, NY005p, OH002p, OH003p, OH005b, OH006p, OH007p, OH008p, OH010p, OK001p, OR011p, OR014p, PA001p, PA002p, PA005p, PA008s, SC001p, SC002p, SC004b, TN012p, TN013p, TN025b, UT002p, VA002p, VA003p, WI002p, WI003p, WI010p, WI013p

Rudbeckia laciniata **Coneflower, cutleaf**

AR005p, CO005p, CO006s, CT003p, CT004p, DE002p, FL012p, IL001s, IL002s, IL004b, IN003p, KY002p, MN004b, MN009b, NC004p, NC005p, NJ001p, NY002p, OH002p, OH007p, OR014p, PA008s, PA010b, SC001p, TN025p, VA003p, WI009s

Rudbeckia nitida **Coneflower, shiny**

CT002p, FL012p, IL004p, MN002p, MN006p, MN010p, NE011p, NY002p, OH007p, OR004p, OR011p, SC003p, VA003p, WI010p

Ruellia brittoniana **Wild petunia, Britton's**

FL012p, FL015p, FL016p, OK001p, OR014p, SC003p

Ruellia malacosperma **Wild petunia, softseed**

OK001p

Rumex acetosella **Sorrel, sheep**

IL002s, NY004s

Rumex altissimus **Dock, pale**

MN009s, PA010b

Rumex crispus **Dock, curly**

IL002s, NY004s, PA008s

Rumex orbiculatus **Dock, greater water**

WI008s

Rumex verticillatus **Dock, swamp**

MN009s, PA008s

Sabal minor **Palmetto, dwarf**

FL004p, FL005p, FL014p, FL015p, LA007p, LA008s, SC003p, VA005p

Sabatia bartramii **Rose gentian, Bartram's**

NC005p

Sabatia calycina **Rose gentian, coastal**

FL015p

Sabatia dodecandra **Rose gentian, marsh**

SC003p

Sabatia kennedyana **Rose gentian, Plymouth**
NC004p, NC005p

Saccharum alopecuroidum **Plumegrass, woolly**
KY002p

Saccharum baldwinii **Plumegrass, narrow**
TN025b

Saccharum giganteum **Plumegrass, sugarcane**
AR004p, TN025b

Saccharum ravennae **Ravennagrass**
CA004s, CO001p, CO003p, CO005p, CT002p, CT004p, GA001p, IL004p, IN001p, MA004p, MD003p, MI001p, MI004p, MN010p, NC008p, NE011p, NY002p, OH001p, OH003p, OH005p, OH007p, OH008p, OK001p, OR008p, OR014p, TN012p, VA003p, WI010p

Sagittaria graminea **Arrowhead, grassy**
CT009p

Sagittaria lancifolia **Arrowhead, bulltongue**
FL005p, FL016p, FL017p, MD001p, NC003p, PA010p

Sagittaria latifolia **Arrowhead broadleaf**
AR005p, CA007p, CA017b, CO004p, CO005p, CT004p, CT009b, FL005p, FL016p, GA003p, IA003s, IL002s, IN003p, KY002p, MA002p, MA003p, MD001p, MD002p, MN004b, MN009s, MO011p, MS003p, MT001p, NC003p, NC005p, NC008p, NJ006p, NJ008p, OH008p, OR008p, OR009p, PA004p, PA008s, PA009p, PA010p, PA012p, SC001p, VA001p, VA004p, VA005p, WI001p, WI007p, WI008s, WI012b

Sagittaria montevidensis **Arrowhead, giant**
MD001p, NC003p

Sagittaria rigida **Arrowhead, sessilefruit**
IL002s, MO011p, PA010p, WI001p, WI007p

Sagittaria sanfordii **Arrowhead, valley**
CA007p

Salicornia virginica **Glasswort, Virginia**
CA007p, CA009p, CA013p, CA017b

Salix alba **Willow, white**
CO003p, CO005p, CT001p, DE001p, DE002p, IA004p, ID007p, IL003p, IL006p, MI002p, MN002p, MN003p, MN006p, MO002p, MT002p, MT005p, MT006p, MT007p, MT015p, NC007p, NJ004p, NV001p, NY003p, OH003p, OH004p, OH008p, OH009p, OK001p, OK004p, OK005p, OR001p, OR005p, OR009p, OR014p, SC002p, SC003p, SD003p, TN011p, TN018p, TN020p, TN021p, TN022p, UT002p, UT003p, VA002p, VA005p, WI002p, WI003p

Salix amygdaloides **Willow, peachleaf**
AR004p, CO003p, CO004p, CO005p, MN009b, MT001p, PA008s, PA010p, UT003p

Salix arctica **Willow, arctic**
MT005p

Salix babylonica **Willow, weeping**
AL002p, AR002p, CA003p, CO005p, DE002p, FL005p, FL012p, FL014p, FL016p, ID003p, IL005p, LA001p, LA007p, MS002p, NC002p, NY003p, OK001p, OK004p, OR005p, OR008p, OR009p, OR011p, OR014p, TN001p, TN002p, TN003p, TN004p, TN006p, TN007p, TN009p, TN010p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN020p, TN022p, TN024p, TX008p, UT002p, VA002p, VA005p

Salix bebbiana **Willow, Bebb**
CO005p, ID001p, MN009p, MT001p, OR014p

Salix boothii **Willow, Booth's**
MT001p, OR014p, UT003p

Salix brachycarpa **Willow, shortfruit**
CO005p, OR014p

Salix caprea **Willow, goat**
CT004p, CT005p, ID007p, IL005p, IL006p, IN001p, MN002p, MN003p, MN006p, MN014p, MT007p, NC007p, NY005p, NY006p, OH003p, OH008p, OK001p, OK004p, OK005p, OR003p, OR005p, OR006p, OR011p, OR014p, OR016p, SC002p, SD003p, TN003p, TN005p, TN007p, TN020p, UT002p, VA005p, WI002p, WI003p

Salix caroliniana **Willow, coastal plain**
AR004p, FL016p

Salix commutata **Willow, undergreen**
OR014p

Salix cordata **Willow, heartleaf**
MN009p

Salix discolor **Willow, pussy**
AL002p, AR002p, CO001p, CO003p, CO005p, DE002p, IA002p, LA003p, MA003p, MN002p, MN009p, MN020p, MT005p, MT015p, NJ005p, NJ008p, NY003p, NY006p, OH003p, OH008p, OH009p, OR008p, OR009p, PA002p, PA008s, PA012p, TN001p, TN002p, TN003p, TN004p, TN006p, TN007p, TN009p, TN017p, TN018p, TN019p, TN021p, TN022p, TN024p, VA004p, VA005p, WI003p

Salix drummondiana **Willow, Drummond's**
MT001p

Salix eriocephala **Willow, Missouri River**
MT007p, OR014p, PA008s

Salix exigua **Willow, narrowleaf**
AR004p, CA002p, CA005p, CO001p, CO003p, CO004p, CO005p, ID001p, ID003p, MA003p, MT001p, MT007p, OR014p, PA008s, PA009p, PA010p, PA012p, UT003p, WA003p, WI012p

Salix geyeriana **Willow, Geyer's**
CO005p, CO005p, MT001p, OR014p

Salix gooddingii **Willow, Goodding's**
CA002p, CA005p, CA007p

Salix hookeriana **Willow, snowbed**
CA017p, MT007p, OR009p, OR014p

Salix irrorata **Willow, deweystem**
CO001p, CO003p, CO005p, OR014p

Salix laevigata **Willow, red**
CA002p, CA005p, CA006p, CA007p, CA009p

Salix lasiolepis **Willow, arroyo**
CA002p, CA005p, CA006p, CA007p, CA009p, CA013p, CA017p, OR009p, OR014p

Salix lucida **Willow, shining**
MN009p, OR014p, PA008s, PA010p

Salix lucida ssp. lasiandra **Willow, Pacific**
CA002p, CA005p, CA007p, CA012p, CA017p, MT001p, OR008p, OR009p, OR014p, WA003p

Salix lutea **Willow, yellow**
MT001p

Salix melanopsis **Willow, dusky**
CA002p, CA017p, OR008p, OR009p, OR014p

Salix monticola **Willow, park**
CO001p, CO003p, CO004p, CO005p, MT001p

Salix nigra **Willow, black**
AR002p, MA003p, MD002p, MN009p, NJ008p, OR014p, PA004p, PA005p, PA008s, PA009p, PA010p, PA012p, SC003p, TN007p, TN017p, TN018p, TN022p, VA004p, VA005p

Salix petiolaris **Willow, meadow**
OR014p

Salix planifolia **Willow, diamondleaf**
CO004p, CO005p

Salix polaris **Willow, polar**
CA003p

Salix purpurea **Willow, purpleosier**
AR004p, CA003p, CO001p, CO003p, CO005p, CT004p, IA002p, ID001p, ID003p, MA003p, MD002p, MI002p, MI003p, MN002p, MN006p, MT007p, NH002p, NJ008p, NY003p, NY005p, NY006p, OH002p, OH003p, OH008p, OR005p, OR009p, OR011p, OR014p, OR016p, PA004p, PA005p, PA008s, PA009p, PA010p, PA013p, SC002p, UT002p, VA004p, VA005p, WI003p, WI012p

Salix sericea **Willow, silky**
MD002p, MN009p, PA008s

Salix sitchensis **Willow, Sitka**
CA009p, OR008p, OR009p, OR014p

Salix wolfii **Willow, Wolf's**
CO005p, OR014p

Salvia lyrata **Sage, lyreleaf**
FL015p, ID002s, IL001s, KY003p, LA002p, NC005p, TN003p, TN013p, TN017p, TN019p, TN022p, WV001p

Salvia penstemonoides **Sage, big red**
AR005p

Sambucus nigra* ssp. *canadensis **Elderberry, blue**
CO001p, CO003p, CO005p, CT004p, DE002p, FL013p, IA002p, IA004p, IL004b, IL005p, KY002p, LA002p, LA008s, MA001s, MA003p, MA004p, MD002p, MO004p, MN002p, MN003p, MN009p, MN014p, MT002p, MT005p, MT006p, MT007p, NC004p, NC005p, NJ001p, NJ002p, NJ005p, NJ008p, NY004s, NY005p, NY006p, OH008p, OR011p, OR014p, PA004p, PA005p, PA006p, PA008s, PA009p, PA010p, PA012p, SD003p, TN003p, TN007p, TN017p, TN018p, TN022p, TN024p, UT002p, VA001p, VA004p, VA005p, WI002s, WI003p

Sambucus racemosa* var. *melanocarpa **Elderberry, black**
CA002p, ID002s, MT001p, OR014p, WA003p

Sanguisorba canadensis **Burnet, Canadian**
CT002p, IL004p, MN009p, NC005p, NJ001p, NY005p, WI010p

Sanguisorba officinalis **Burnet, western**
MN010p, NY004s, OR004p, OR016p

Sarracenia alata **Trumpets, yellow**
NY001b

Sarracenia flava **Pitcherplant, yellow**
CA004s, FL005p, FL016p, NY001b

Sarracenia leucophylla **Pitcherplant, crimson**
CA004s, FL005p, NC004p, NY001b

Sarracenia minor **Pitcherplant, hooded**
CA004s, NY001b

Sarracenia psittacina **Pitcherplant, parrot**
FL005p, FL016p, NY001b

Sarracenia purpurea **Pitcherplant, purple**
FL005p, FL016p, MN001p, NJ001p, NJ007s, NY001b, PA008s, PA010p, WI012p

Sarracenia rubra **Pitcherplant, sweet**
FL005p, NY001b

Saururus cernuus **Tail, lizard's**
AR004p, AR005p, FL016p, FL017p, KY002p, LA002p, MD001p, MD002p, NC003p, NC005p, NC008p, NJ005p, NJ008p, OH008p, PA004p, PA008s, PA009p, PA010p, PA012p, VA001p, VA004p, VA005p, WI001p, WI012p

Saxifraga pensylvanica **Saxifrage, eastern swamp**
IL004p, MN001p, MN004b, MN009b

Schinus terebinthifolius **Brazilian peppertree**
CA004s

Schoenoplectus acutus* var. *acutus **Bulrush, hardstem**
CA007p, CA010s, CA017b, CO004p, CO005p, CT002p, GA002p, IA003s, IL002s, IN003p, MA003p, MN009s, MT001p, NJ008p, OK003p, OR008p, OR009p, PA004p, PA008s, PA010b, PA012p, UT001s, VA004p, WI001p, WI007p, WI012b

Schoenoplectus americanus **Bulrush, chairmaker's**
CO003p, CO004p, CO005p, FL005p, FL016p, MA002p, MT001p, NJ008p, OK003p, OR009p, PA004p, PA008s, PA010b, PA012p, UT001s, VA001p, VA004p, WI001p, WI007p, WI012p

Schoenoplectus californicus **Bulrush, California**
AL003p, CA010s, CA013p, CA017b, CO004p, FL005p, FL016p, FL017p, LA006p, MS003p, PA010p

Schoenoplectus fluviatilis **Bulrush, river**
CT009p, IL002s, IN003p, MA002p, MA003p, MD002p, MN009s, NJ008p, PA008s, PA010p, PA012p, VA004p, WI001p, WI007p, WI012p

Schoenoplectus maritimus **Bulrush, cosmoplitan (saltmarsh)**
CA017b, CO004p, MT001p, PA008s, PA010p, UT001s, WI001s

Schoenoplectus pungens var. pungens **Common threesquare**
CA017b, CT009p, IN003p, MA003p, MD002p, MT001p, NJ005p, NJ008p, PA009p, PA010b, VA004p, VA005p, WI001p

Schoenoplectus robustus **Bulrush, sturdy**
CA002p, CA007p, CA009p, CA010s, CA013p, CA017b, CT009p, FL017p, MA002p, MD002p, NJ008p, PA010b, VA001p, VA004p

Schoenoplectus tabernaemontani **Bulrush, softstem**
CA007p, CO004p, CO005p, CT002p, CT009b, FL005p, FL016p, FL017p, GA002p, IN003p, KY002p, MA002p, MA003p, MD002p, MN004b, MN009b, MT001p, NC008p, NJ005p, NJ008p, NY005p, OH008p, OR008p, OR009p, PA004p, PA008s, PA009p, PA010p, PA012p, SC001p, VA001p, VA004p, VA005p, WI001p, WI007p, WI008s, WI012b

Schoenoplectus torreyi **Bulrush, Torrey's**
PA008s

Scirpus atrovirens **Bulrush, green**
CT009b, IL004b, IN003p, KY002p, MA002p, MA003p, MN004b, MN009b, MN013s, MT001p, NC001p, NC005p, NJ008p, PA004p, PA008s, PA009p, PA010b, PA012p, VA001p, VA004p, WI001p, WI007p, WI008s, WI012b

Scirpus cernuus **Bulrush, low**
CA009p

Scirpus cyperinus **Woolgrass**
CT009b, FL005p, FL016p, IN003p, LA002p, MA002p, MA003p, MD002p, MN004b, MN009b, MS006p, MS007p, MS009p, NC003p, NC005p, NJ005p, NJ008p, PA004p, PA008s, PA009p, PA010b, PA012p, TN026p, VA001p, VA004p, VA005p, WI001p, WI007p, WI008s, WI012b

Scirpus expansus **Bulrush, woodland**
CT002p, CT009b

Scirpus georgianus **Bulrush, Georgia**
NJ005p

Scirpus hattorianus **Bulrush, mosquito**
CT009s

Scirpus microcarpus **Bulrush, panicled**
CA017b, CO003p, CO004p, CO005p, ID002s, MN009s, MT001p, OR008p, OR009p, PA008s, PA010p

Scirpus nevadensis **Bulrush, Nevada**
CA017b

Scirpus pallidus **Bulrush, cloaked**
CO004p, CO005p

Scirpus polyphyllus **Bulrush, leafy**
NJ005p, PA008s

Scutellaria integrifolia **Helmet flower**
AR005p, FL015p, NC005p

Scutellaria lateriflora **Skullcap, blue**
AR005p, IL004p, KY003p, ME001s, MN009s, NE003s, WV001p

Selaginella apoda **Spikemoss, meadow**
NC005p

Selaginella uncinata **Spikemoss, blue**
AR005p, CT002p, NC004p, NC005p, SC002p, SC003p

Senecio aureus **Ragwort, golden**
CT002p, KY003p, MN004b, MN009s, NJ001p, PA008s, PA010p, SC003p, TN013p

Senecio clevelandii **Ragwort, Cleveland's**
CA002p

Senecio pauperculus **Groundsel, balsam**
MN004b, MN009s

Senecio triangularis **Ragwort, arrowleaf**
CO005p

Senna hebecarpa **Senna, American**
IL004b, IN003p, KY003p, MN009b, MN010p, NC005p, NE011p, NY004s, OH007p, OH008p, OR014p, WI008s, WI009s

Senna marilandica **Senna, Maryland**
AR004p, KY002p, MN009b, NC005p, NE003s, SC004s, WI010p

Sesbania drummondii **Rattle-bush, Drummond's**
SC003p

Sesbania herbacea **Bigpod, sesbania**
AR003s, GA002s, GA003s, MO011s, TX002s, WI001s, WI007s

Sesuvium portulacastrum **Shoreline seapurslane**
FL006p, FL011p

Sidalcea candida **Checkerbloom, white**
CT003p, GA001p, OR014p

Sidalcea neomexicana **Checkerbloom, salt spring**
CA002p

Sidalcea oregana **Checkerbloom, Oregon**
GA001p, MN010p, OR004p

Sideroxylon lycioides **Buckthorn bully**
FL015p, SC003p

Silene nivea **Campion, evening**
MN009p

Silphium perfoliatum **Cup plant**
AR005p, IA003s, IL001s, IL004b, IN003p, KY002p, MA004p, MN004b, MN009b, MN011s, NE011p, OR014p, PA008s, SC003p, TN025p, WI008b, WI009b, WI010p

Sisyrinchium angustifolium **Blue-eyed grass, narrowleaf**
AR005p, CO005p, CT002p, ID002s, KY002p, LA002p, MA004p, MN009s, NC005p, NC006p, NJ007s, OH002p, OH005p, OR004p, OR016p, TN017p, TN025p

Sisyrinchium atlanticum **Blue-eyed grass, eastern**
FL008p, FL015p, NC005p, SC003p

Sisyrinchium bellum **Blue-eyed grass, western**
CA002p, CA004s, CA005p, CA006p, CA007p, CA009p, CA010s, CA011s, CA013p, CA017b, CO006s, NC006p, NE011p, NY002p, OH005p, OR004p, OR014p, OR016p, UT001s

Sisyrinchium californicum **Blue-eyed grass, golden**
CA002p, CA005p, CA006p, CA007p, CA009p, CA013p, CA017b, NC005p, OR004p, OR014p

Sisyrinchium idahoense **Blue-eyed grass, Idaho**
NC005p, NE011p

Sium suave **Waterparsnip, hemlock**
AR005p, IL004b

Smilax walteri **Greenbrier, coral**
SC003p

Solidago gigantea **Goldenrod, giant**
NC005p, NJ005p, PA008s

Solidago latissimifolia **Goldenrod, Elliott's**
NJ005p

Solidago patula **Goldenrod, roundleaf**
IL004b, IN003p, NC005p, PA008s

Solidago sempervirens **Goldenrod, seaside**
LA002p, MD002p, NJ001p, NJ005p, NJ008p, PA012p, TN025b, VA004p

Solidago spectabilis* var. *confinis **Goldenrod, Nevada**
CA002p

Solidago stricta **Goldenrod, wand**
FL014p, NC005p, NJ005p

Solidago uliginosa **Goldenrod, bog**
PA008s, PA012p

Sparganium americanum **Bur-reed, American**
KY002p, MA003p, MD002p, NJ008p, PA008s, PA009p, PA010p, PA012p, VA004p, VA005p

Sparganium androcladum **Bur-reed, branched**
CT009p

Sparganium angustifolium **Bur-reed, narrowleaf**
CA017b, OR009p

Sparganium erectum **Bur-reed, simplestemmed**
CA017b

Sparganium eurycarpum **Bur-reed, giant (broadfruit)**
CO004p, CO005p, CT009p, GA002p, IL002s, IN003p, MD002p, MN004b, MN009s, OR008p, PA004p, PA008s, PA010p, PA012p, VA005p, WI001p, WI007p, WI012b

Sparganium fluctuans **Bur-reed, floating**
WI012p

Spartina alterniflora **Cordgrass, smooth**
FL005p, FL016p, LA006p, LA009p, MA002p, MA003p, MD002p, NJ008p, NJ009p, PA009p, VA001p, VA004p, VA005p

Spartina bakeri **Cordgrass, sand**
FL003p, FL005p, FL006p, FL008p, FL010p, FL016p, FL017p, NC008p, SC001p, VA005p

Spartina cynosuroides **Cordgrass, big**
MD002p, NJ008p, PA004p, VA001p, VA004p

Spartina foliosa **Cordgrass, California**
CA017b

Spartina gracilis **Cordgrass, alkali**
CO005p, MT001p

Spartina patens **Cordgrass, saltmeadow**
FL005p, FL006p, FL010p, FL011p, FL016p, FL017p, GA004p, LA006p, MA002p, MA003p, MD002p, MD003p, MJ009p, NC009p, NJ008p, NJ009p, NJ010p, PA004p, PA009p, SC001p, VA001p, VA004p, VA005p

Spartina pectinata **Cordgrass, prairie**
CO004p, CO005p, CT002p, CT004p, IA005s, IL004p, IN003p, KY002p, MA002p, MA004p, MN004b, MN007s, MN009b, MN011s, MO001s, MO002p, MO009s, MT001p, NC008p, NE007s, NE011p, NJ005p, NJ008p, NY004s, OH007p, OH008p, OR014p, PA004p, PA008s, PA010p, VA003p, VA004p, WI001p, WI006p, WI007p, Wi008b, WI009p, WY001s, WI012b

Spartina spartinae **Cordgrass, gulf**
FL016p, TX004p

Spiraea alba **Meadowsweet, white**
IL004p, MN001p, MN009b, PA008s, PA012p, WI012p

Spiraea alba* var. *latifolia **Meadowsweet, white**
MA003p, NJ001p, NJ005p, NJ008p, PA004p, VA004p

Spiraea douglasii **Spirea, rose**
CA002p, CA007p, CA012p, CA017b, ID001p, ID002s, ID003p, MA001s, MT001p, OR008p, OR009p, OR014p, WA003p

Spiraea tomentosa **Steeplebush**
CT009b, KY002p, MN004b, MN009b, NJ001p, NJ005p, NJ008p, NY004s, PA004p, PA008s, PA010p, VA004p, WI008s

Spiranthes cernua **Ladies'-tresses, nodding**
FL015p, NC005p, NE011p

Spiranthes odorata **Ladies'-tresses, marsh**
NC004p, NY005p

Spiranthes vernalis **Ladies'-tresses, spring**
FL015p

Spirodela polyrrhiza **Duckweed, greater**
IL002s

Sporobolus virginicus **Dropseed, seashore**
FL016p

Stachys ajugoides **Hedgenettle, bugle**
CA013p

Stachys albens **Hedgenettle, whitestem**
CA002p

Stachys chamissonis **Hedgenettle, coastal**
CA002p

Stachys palustris* ssp. *pilosa **Hedgenettle, marsh**
CA012p

Stachys pycnantha **Hedgenettle, shortspike**
CA002p

Stuckenia pectinatus **Pondweed, sago**
CA017b, CO004p, CO005p, GA002p, IL002s, MA003p, MO011p, PA004p, PA010p, PA012p, WI001b, WI007p, WI012b

Styrax americanus **Snowbell, American**
CA004s, CT005p, FL014p, FL015p, LA002p, MA001s, NC005p, NC007p, NE003s, NY004s, SC003p, VA005p

Suaeda californica **Seablite, California**
CA002p, CA007p

Symplocarpus foetidus **Skunk cabbage**
MA003p, MN009p, PA010p, WI012p

Tamarix gallica **Tamarisk, French**
MA001s, MT005p

Tamarix parviflora **Tamarisk, smallflower**
NV001p, OR011p

Tamarix ramosissima **Saltcedar**
CO003p, CO005p, CT004p, IA002p, IA004p, MN002p, MN006p, MN014p, OH008p, OR005p, OR011p, OR014p, SC002p, UT002p, WI002s, WI003p

Taraxacum officinale **Dandelion, common**
IL002s, ME001s

Taxodium ascendens **Cypress, pond**
CA003p, CA004s, CT005p, FL001p, FL003p, FL005p, FL007p, FL009p, FL010p, FL014p, FL015p, FL016p, LA002p, MO005p, NJ004p, NY004s, OR011p, OR014p, OR016p, SC003p, VA005p

Taxodium distichum **Cypress, bald**
AL007p, AR001p, AR002p, CA003p, CA004s, CT004p, CT005p, DE002p, FL001p, FL003p, FL004p, FL005p, FL007p, FL008p, FL009p, FL010p, FL012p, FL013p, FL014p, FL015p, FL016p, FL017p, GA002p, IA001p, IN001p, IN002p, LA001p, LA002p, LA003p, LA004p, LA005p, LA007p, LA008s, MA001s, MD002p, MI003p, MI004p, MN002p, MO002p, MO005p, MS001p, MS002p, MT007p, NC001p, NC002p, NC005p, NJ004p, NJ008p, NY003p, NY004s, NY005p, OH001p, OH003p, OH008p, OK001p, OK004p, OK005p, OR003p, OR011p, OR014p, OR015p, OR016p, PA003p, PA004p, PA005p, PA010p, PA012p, SC003p, TN001p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN008p, TN009p, TN011p, TN012p, TN017p, TN018p, TN019p, TN020p, TN021p, TN022p, TN024p, VA002p, VA004p, VA005p

Taxodium mucronatum **Cypress, Montezuma bald**
CA003p, CA004s, OR011p

Teucrium canadense **Germander, Canada**
AR005p, CO001p, CT002p, MI001p, MN009s, MN010p, NC004p, NE011p, NY004s, OH002p, OH007p, WV001p

Thalia dealbata **Alligator-flag, powdery**
AR005p, MA004p, MD001p, MS006p, MS007p, MS008p, MS009p, NC003p, NC005p, NC008p, SC001p, SC003p, TN026p

Thalia geniculata **Alligator-flag, bent**
FL010p, FL016p, FL017p, MD001p

Thalictrum alpinum **Meadow-rue, alpine**
CA006p

Thalictrum clavatum **Meadow-rue, mountain**
NC005p

Thalictrum dasycarpum **Meadow-rue, purple**
AR005p, IL004b, MN001p, MN004b, MN009b, MN011s, NC005p, NE003s, OR004p, WI008b, WI009p

Thalictrum dioicum **Meadow-rue, early**
IL004b, MN001p, MN004b, MN009b, OR004p, PA008s, WI006p, WI008b

Thalictrum pubescens **King of the meadow**
PA008s

Thelypteris kunthii **Fern, Kunth's maiden**
FL009p, FL012p, LA001p, NC004p, OR011p, SC001p, SC003p

Thelypteris noveboracensis **Fern, New York**
MA004p, PA004p, PA010p, SC003p

Thelypteris palustris* var. *pubescens **Fern, eastern marsh**
MN001p, OR004p

Thuja occidentalis **Cedar, northern white**
AR002p, CA003p, CA004s, CO005p, CT001p, CT005p, CT006p, DE001p, DE002p, IA001p, IA004p, ID003p, IN001p, MA001s, MI002p, MI003p, MI004p, MN002p, MN003p, MN004b, MN006p, MN009p, MN020p, MT015p, MN017p, MT005p, MT006p, MT007p, NC002p, NC007p, NJ002p, NJ004p, NY003p, NY004s, NY005p, OH001p, OH002p, OH003p, OH004p, OH008p, OH009p, OK001p, OK005p, OR006p, OR008p, OR009p, OR010p, OR011p, OR014p, PA001p, PA003p, PA005p, PA006p, PA007p, PA011p, SC002p, TN002p, TN003p, TN004p, TN006p, TN007p, TN012p, TN014p, TN016p, TN018p, TN022p, TN024p, UT002p, VA002p, VA005p, WI002s, WI003p, WI004p

Tolmiea menziesii **Youth on age**
CA017b, CT002p, NY005p, OR004p, OR014p

Toxicodendron vernix **Sumac, poison**
PA008s

Tradescantia occidentalis **Spiderwort, prairie**
CO003p, MN004b, MN009b, MN013s

Tradescantia ohiensis **Bluejacket**
AR005p, FL008p, IL004b, KY002b, MN007s, MN009b, MN013s, MO003b, NC005p, NE003s, NE011p, PA008s, WI008b, WI009b, WI012b

Triadenum virginicum **St. Johnswort, Virginia marsh**
CT009s, PA008s

Trifolium fragiferum **Clover, strawberry**
CA010s, CA014s, CA015s, CA016s, CO007s, ID006s, NM001s, UT001s, UT004s, UT005s

Trifolium microcephalum **Clover, smallhead**
CA010s

Trifolium willdenowii **Clover, tomcat**
CA010s

Triglochin maritimum **Arrowgrass, seaside**
CA009p, CA017b, CO004p, PA010p

Trillium cernuum **Trillium, nodding**
KY002p, MN009p, NY004s, NY005p, TN015p, TN017p, TN019p, TN023p

Trillium pusillum **Dwarf wakerobin**
NY005p, TN015p, TN019p, TN023p

Tripsacum dactyloides **Gamagrass, eastern**
CO009s, GA003s, FL003p, FL005p, FL008p, FL009p, FL010p, FL014p, FL016p, IA003s, KS001s, KS002s, KS004s, KY002b, MA004p, MN009s, MO001s, MO006s, MO009s, MO010s, NE001s, NE002s, NJ005p, OK002s, OK003p, OK006s, PA008s, TX002s, TX011s

Trisetum spicatum **False-oats, spiked**
MT001p

Tsuga canadensis **Hemlock, eastern**
AL001p, CA004s, CT001p, CT004p, CT005p, DE001p, DE002p, ID002s, IL005p, IN001p, MA001s, MA003p, ME002p, MI002p, MI003p, MI004p, MI005p, MN002p, MN003p, MN006p, MN014p, MN017p, MN020p, NC007p, NY003p, NY004s, NY005p, OH001p, OH003p, OH008p, OH009p, OR006p, OR009p, OR011p, OR014p, PA001p, PA005p, PA006p, PA007p, PA010p, PA011p, PA012p, SC002p, TN002p, TN003p, TN004p, TN005p, TN006p, TN007p, TN009p, TN011p, TN012p, TN016p, TN017p, TN018p, TN019p, TN022p, TN024p, UT002p, VA005p, WI003p, WI004p

Typha angustifolia **Cattail, narrowleaf**
CA007p, CA017b, CO005p, CT002p, CT009p, FL005p, MA002p, MA003p, MD001p, MD002p, MT001p, NC003p, NJ005p, NJ008p, OH008p, OR011p, PA004p, PA008s, PA009p, PA010b, PA012p, VA001p, VA004p, VA005p, WI001p, WI007p, WI012b

Typha domingensis **Cattail, southern**
CO004p, PA010p

Typha latifolia **Cattail, broadleaf**
AR005p, CA007p, CA010s, CA013p, CA017b, CO004p, CO005p, CT002p, CT009p, FL016p, IL001s, IL002s, KY002p, MA002p, MA003p, MD001p, MD002p, MN009p, MT001p, NC003p, NC008p, NJ005p, NJ006p, NJ008p, OR008p, OR009p, PA004p, PA008s, PA009p, PA010b, PA012p, UT001s, UT003p, VA001p, VA004p, VA005p, WI001p, WI007p, WI012b

Ulmus americana ***Elm, American***
CA004s, FL001p, FL005p, FL009p, FL014p, FL016p, LA008s, MA001s, MD002p, MN002p, MO002p, MT005p, MT006p, NJ004p, NJ005p, NY004s, OH003p, TN017p, TX013p, VA005p

Urtica dioica **Nettle, stinging**
CA010s, IL002s, KY003s, MA004p, ME001s

Utricularia macrorhiza **Bladderwort, common**
PA010p

Vaccinium corymbosum **Blueberry, highbush**
CT001p, CT005p, CT006p, CT008p, DE002p, IA004p, IL003p, IL004p, IL005p, IL006p, FL001p, FL014p, FL015p, GA001p, MA001s, MA003p, MD002p, MI001p, MI002p, MI005p, MN002p, MN014p, MN020p, MO004p, NJ001p, NJ008p, NY003p, NY004s, NY006p, OH001p, OH008p, OK005p, OR011p, PA002p, PA004p, PA006p, PA009p, PA010p, PA012p, SC001p, SD003p, TN003p, TN010p, VA004p, VA005p, WI002p

Vaccinium elliottii **Blueberry, Elliott's**
FL015p

Vaccinium macrocarpon **Cranberry, large**
GA001p, IA004p, ID001p, MA001s, MA003p, MA004p, NC005p, NJ001p, NJ005p, NY004s, NY005p, OH003p, OR002p, OR014p, SC002p, SD003p, TN013p

Vaccinium myrtilloides **Blueberry, velvetleaf**
MA001s, OR014p

Vaccinium oxycoccos **Cranberry, small**
CA003p, OR011p, OR014p

Vaccinium uliginosum **Blueberry, bog**
NY004s, OR002p

Valeriana edulis **Tobacco root**
MN009b

Vallisneria americana **American eelgrass**
CT009p, GA002p, GA003p, MA003p, MO011p, NC003p, PA004p, PA010p, PA012p, WI001b, WI007p, WI012p

Veratrum viride **False hellebore, green**
MT001p, PA008s, PA010p

Verbena bonariensis **Vervain, purpletop**
CT002p, CT004p, CT008s, FL012p, LA002p, ME001s, ME003s, NC004p, NE003s, NJ007s, NY002p, NY005p, OH005b, OR011p, OR014p, SC001p, SC004b, VA003p

Verbena hastata **Vervain, blue**
CO004p, CO005p, CT009s, IA003s, IL001s, IL004b, KY002b, MA003p, MA004p, MD002p, ME001s, MN004b, MN007s, MN009b, MN011s, NJ005p, NJ008p, PA004p, PA008s, PA009p, PA010b, VA004p, WI007p, WI008s, WI009b, WI010p, WI012b

Verbesina alternifolia **Wingstem**
AR005p, CT002p, PA008s

Vernonia arkansana **Ironweed, bur**
AR005p, TN025b, OR014p

Vernonia baldwinii **Ironweed, Baldwin's**
AR005p

Vernonia fasciculata **Ironweed, prairie**
AR005p, CT002p, CT004p, IL004b, MN007s, MN004b, MN009b, MN011s, NJ005p, OR014p, WI008b, WI009b, WI010p, WI012p

Vernonia missurica **Ironweed, Missouri**
MN009s, NC004p

Vernonia noveboracensis **Ironweed, New York**
CT002p, CT004p, MA003p, MD002p, MN006p, MN009p, MN010p, NC003p, NC004p, NC006p, NJ001p, NJ005p, NJ008p, NY004s, OR014p, PA004p, PA008s, PA009p, PA010p, PA012p, VA004p, VA005p

Veronica americana **Speedwell, American**
CA017b, CO005p, MT001p

Veronica peregrina **Speedwell, hairy purslane**
IL002s

Veronica wormskjoldii **Speedwell, American alpine**
OR014p

Veronicastrum virginicum **Culver's root**
AR005p, CO003p, CT002p, GA001p, IA003s, IL001s, IL004b, IN003p, KY002p, MA004p, MN004b, MN009b, MN010p, NC006p, NE003s, NE011p, NJ001p, NY005p, OR011p, OR014p, PA008s, SC001p, SC003p, WI006p, WI008b, WI009b, WI010p, WI012b, WI013p

Viburnum dentatum* var. *dentatum **Arrowwood, southern**
SC003p

Viburnum dentatum* var. *lucidum **Arrowwood, northern**
NY004s, PA008s, PA010p, PA012p

Viburnum edule **Squashberry**
ID002s

Viburnum lentago **Nannyberry**
CO001p, CO003p, CO005p, DE002p, IA002p, IN001p, KY002p, MA001s, MA003p, MA004p, MD002p, MN002p, MN003p, MN006p, MN009p, MO002p, MT002p, MT005p, MT006p, MT007p, MT015p, NE003s, NJ004p, NJ005p, NJ008p, NY003p, NY004s, NY006p, OH008p, OR005p, OR011p, OR014p, PA004p, PA005p, PA008s, PA009p, PA010p, PA012p, TN003p, TN017p, VA004p, VA005p, WI003p, WI006p

Viburnum nudum **Possumhaw**
CT005p, FL014p, FL015p, KY002p, LA002p, MD002p, MN002p, NC005p, NJ001p, NY005p, OR011p, OR014p, PA001p, PA012p, SC003p, VA001p

Viburnum nudum* var. *cassinoides **Withe-rod**
CT005p, MA003p, MA004p, MN006p, MN009p, NY003p, NY004s, NY005p, OR014p, PA008s, PA012p

Viburnum obovatum — **Arrowwood, small-leaf**
FL003p, FL005p, FL009p, FL010p, FL012p, FL014p, FL015p, FL016p, LA008s, NC007p, SC003p

Viburnum opulus var. americanum — **Cranberrybush, American**
CO001p, CO003p, CO005p, CT004p, CT006p, DE001p, IA001p, IA002p, IA004p, IN001p, MA001s, MA003p, MA004p, MI002p, MI003p, MI004p, MI005p, MN002p, MN003p, MN006p, MN009p, MN014p, MO002p, MT002p, MT005p, MT006p, MT007p, MT015p, NJ001p, NJ002p, NJ004p, NJ008p, NY003p, NY004s, NY005p, NY006p, OH003p, OH008p, OH009p, OK005p, OR005p, OR008p, OR014p, PA004p, PA005p, PA007p, PA008s, PA009p, PA010p, PA011p, PA012p, SD003p, TN003p, TN013p, UT002p, VA004p, VA005p, WI002s, WI003p, WI004p, WI006p, WI012p

Vicia sativa — **Vetch, common**
CA015s, CO007s, IL002s, MT003s

Viola affinis — **Violet, Le Conte sand**
FL012p

Viola blanda — **Violet, sweet white**
KY002p, MN001p, TN015p, TN023p

Viola conspersa — **Violet, American dog**
MN001p, MN009p

Viola cucullata — **Violet, marsh blue**
CT002p, NY005p, OH002p, OH007p, OR004p, WA002p

Viola glabella — **Violet, pioneer**
CA013p

Viola macloskeyi ssp. pallens — **Violet, smooth white**
MA004p, MN001p

Viola missouriensis — **Violet, Le Conte sand**
OR004p

Viola novae-angliae — **Violet, New England blue**
MN001p

Viola pubescens var. pubescens — **Violet, downey yellow**
SC003p, TN003p, TN019p

Viola sagittata — **Violet, arrowleaf**
KY002p, MN004b, MN009b, NY005p, OR004p

Viola septemloba — **Violet, southern coastal**
SC003p

Viola sororia — **Violet, common blue**
KY002p, NY004s, OR014p

Viola striata **Violet, striped cream**
MN009p, NE011p, OR004p

Vitis californica **Grape, California wild**
CA002p, CA006p, CA007p, CA009p, CA013p, CA017b, MT001p, OR014p

Vitis riparia **Grape, riverbank**
ID002s, MN009s, NE011p, NY004s, OR014p

Vitis rotundifolia **Muscadine**
AL002p, AR002p, IL006p, MO004p, TN003p, TN006p, TN007p, TN018p, TX008p

Vulpia myuros **Fescue, rat-tail**
CA010s

Washingtonia filifera **Fan palm, California**
CA004s, MA001s, OK001p, OR014p

Wisteria frutescens **Wisteria, American**
FL012p, MA001s, MN002p, NC005p, NE003s, NY004s, NY005p, OH008p, OR011p, OR014p, SC003p, TN013p, WI010p

Wolffia columbiana **Watermeal, Columbia**
WI012p

Woodwardia areolata **Chainfern, netted**
FL010p, LA003p, NC005p, NY005p, PA010p, SC003p

Woodwardia fimbriata **Chainfern, giant**
CA006p, CA007p, CA013p, CA017b, OR014p

Woodwardia radicans **Chainfern, rooting**
WA001p

Wyethia helianthoides **Mule-ears, sunflower**
ID002s, ID004s

Xanthorhiza simplicissima **Yellowroot**
CT005p, FL014p, NC005p, NY003p, OH008p, OR014p, SC003p, TN010p, TN015p, TN023p

Zantedeschia aethiopica **Calla lily**
AR002p, CA004s, CT004p, GA001p, LA007p, MI005p, OH005p, PA002p, SC001p, SD003p, WI002p, WI011p

Zenobia pulverulenta **Honeycup**
MA001s, NY005p, OR002p, OR011p, OR014p, SC003p

Zephyranthes atamasca **Lily, Atamasco**
FL014p, FL015p, SC003p, TN025p

Zigadenus elegans **Deathcamas, mountain**
MN009p, NY005p

Zingiber zerumbet **Ginger, bitter**
FL012p

Zizania aquatica **Wildrice, annual**
GA002b, GA003s, IL002s, MA004p, MO011b, PA008s, PA010b, PA012p, TX004p, WI001b, WI007s, WI012b

Zizaniopsis miliacea **Wildrice, southern**
AL003p, LA006p, TX004p

Zizia aptera **Meadow zizia**
AR005p, CT002p, IL004b, MN009b, MN011s, NY005p, WI008b, WI009

Directory of Wetland Plant Vendors

Part 3: Alternate Plant Names

Synonym (old name)	**Currently Accepted Name**
Acer dasycarpum	*Acer saccharinum*
Acer drummondii	*Acer rubrum* var. *drummondii*
Agrostis alba	*Agrostis gigantea*
Agrostis palustris	*Agrostis stolonifera*
Agrostis tenuis	*Agrostis capillaris*
Alnus crispa	*Alnus viridis* ssp. *crispa*
Alnus rugosa	*Alnus incana* ssp. *rugosa*
Alnus sinuata	*Alnus viridis* ssp. *sinuata*
Alnus tenuifolia	*Alnus incana* ssp. *tenuifolia*
Anacharis canadensis	*Elodea canadensis*
Aquilegia hinckleyana	*Aquilegia chrysantha var. hinckleyana*
Aquilegia shockleyi	*Aquilegia formosa*
Aster adscendens	*Symphyotrichum ascendens*
Aster ascendens	*Symphyotrichum ascendens*
Aster alpigenus	*Oreostemma alpigenum* var. *alpigenum*
Aster bigelovii	*Machaeranthera bigelovii* var. *bigelovii*
Aster carolinianus	*Ampelaster carolinianus*
Aster lucidulus	*Symphyotrichum puniceus*
Aster simplex	*Symphyotrichum lanceolatum* ssp. *lanceolatum var. interior*
Aster umbellatus	*Doellingeria umbellata*
Aster vimineus	*Symphyotrichum lateriflorum* var. *lateriflorum*
Axonopus affinis	*Axonopus fissifolius*
Baccharis glutinosa	*Baccharis salicifolia*
Baccharis viminea	*Baccharis salicifolia*
Beckmannia eruciformis	*Beckmannia syzigachne*
Betula glandulosa	*Betula nana*
Bidens polylepis	*Bidens aristosa*
Brunnichia cirrhosa	*Brunnichia ovata*
Bumelia lycioides	*Sideroxylon lycioides*
Calamagrostis cinnoides	*Calamagrostis coarctata*
Calamagrostis crassiglumis	*Calamagrostis stricta* ssp. *inexpansa*
Calamagrostis fernaldii	*Calamagrostis stricta* ssp. *inexpansa*
Calamagrostis inexpansa	*Calamagrostis stricta* ssp. *inexpansa*
Cardamine scutata	*Cardamine regeliana*
Carex breviligulata	*Carex densa*
Carex howei	*Carex atlantica* ssp. *capillacea*
Carex limnophila	*Carex microptera*
Carex smalliana	*Carex lonchocarpa*
Cassia hebecarpa	*Senna hebecarpa*
Cassia marilandica	*Senna marilandica*
Castilleja exilis	*Castilleja minor* ssp. *minor*
Castilleja mogollonica	*Castilleja sulphurea*
Celtis reticulata	*Celtis laevigata* var. *reticulata*
Cladium jamaicense	*Cladium mariscus* ssp. *jamaicense*
Cladium leptostachyum	*Cladium mariscus* ssp. *jamaicense*
Colocasia antiquorum	*Colocasia esculenta*
Conoclinium coelestinum	*Eupatorium coelestinum*
Cornus baileyi	*Cornus sericea* ssp. *sericea*
Cornus stolonifera	*Cornus sericea* ssp. *sericea*

Synonym (old name)	**Currently Accepted Name**
Crinum strictum	*Crinum americanum*
Cyperus alternifolius	*Cyperus involucratus*
Cyperus engelmannii	*Cyperus odoratus*
Cyperus ferax	*Cyperus odoratus*
Cyperus ferruginescens	*Cyperus odoratus*
Cyperus stenolepis	*Cyperus strigosus*
Cypripedium calceolus	*Cypripedium parviflorum*
Dichromena colorata	*Rhynchospora colorata*
Dichromena latifolia	*Rhynchospora latifolia*
Dioscorea hirticaulis	*Dioscorea villosa*
Dipsacus sylvestris	*Dipsacus fullonum* ssp. *sylvestris*
Distichlis spicata var. stricta	*Distichlis spicata*
Distichlis stricta	*Distichlis spicata*
Dryopteris atropalustris	*Dryopteris celsa*
Dryopteris dilatata	*Dryopteris expansa*
Dryopteris spinulosa	*Dryopteris carthusiana*
Dryopteris thelypteris	*Thelypteris palustris* var. *pubescens*
Eleocharis coloradoensis	*Eleocharis parvula*
Eleocharis macrostachya	*Eleocharis palustris*
Eleocharis mamillata	*Eleocharis palustris*
Eleocharis tuberosa	*Eleocharis dulcis*
Elodea brandegeeae	*Elodea canadensis*
Elodea linearis	*Elodea canadensis*
Eragrostis campestris	*Eragrostis refracta*
Erianthus alopecuroides	*Saccharum alopecuroidum*
Erianthus giganteus	*Saccharum giganteum*
Erianthus ravennae	*Saccharum ravennae*
Erianthus strictus	*Saccharum baldwinii*
Eupatoriadelphus dubius	*Eupatorium dubium*
Eupatoriadelphus fistulosus	*Eupatorium fistulosum*
Eupatoriadelphus maculatus	*Eupatorium maculatum* var. *maculatum*
Eurystemon mexicanus	*Heteranthera mexicana*
Eustoma grandiflorum	*Eustoma russellianum*
Festuca arundinacea	*Lolium arundinaceum*
Frankenia grandifolia	*Frankenia salina*
Grindelia humilis	*Grindelia hirsutula* var. *hirsutula*
Habranthus texanus	*Habranthus tubispathus*
Heracleum lanatum	*Heracleum maximum*
Hibiscus militaris	*Hibiscus laevis*
Hymenocallis coronaria	*Hymenocallis caroliniana*
Hymenocallis crassifolia	*Hymenocallis floridana*
Hymenocallis eulae	*Hymenocallis liriosome*
Hymenocallis keyensis	*Hymenocallis latifolia*
Hymenocallis kimballiae	*Hymenocallis latifolia*
Hymenocallis laciniata	*Hymenocallis floridana*
Hymenocallis occidentalis	*Hymenocallis caroliniana*
Hypericum apocynifolium	*Hypericum nudiflorum*
Hypoxis leptocarpa	*Hypoxis hirsuta curtissii*
Iris longipetala	*Iris missouriensis*

Synonym (old name)	**Currently Accepted Name**
Juncus platyphyllus	*Juncus dichotomus*
Juniperus silicicola	*Juniperus virginiana* var. *silicicola*
Justicia mortuifluminis	*Justicia americana*
Keysseria erici	*Lagenifera erici*
Keysseria helenae	*Lagenifera helenae*
Kosteletzkya althaeifolia	*Kosteletzkya virginica*
Kosteletzkya smilacifolia	*Kosteletzkya virginica*
Lilium wigginsii	*Lilium pardalinum* ssp. *wigginsii*
Lycopodium lucidulum	*Huperzia lucidula*
Matteuccia pensylvanica	*Matteuccia struthiopteris*
Mazus japonicus	*Mazus pumilus*
Mentha citrata	*Mentha aquatica*
Mimulus nasutus	*Mimulus guttatus*
Myosotis alpestris	*Myosotis asiatica*
Myosotis palustris	*Myosotis scorpioides*
Myrica cerifera	*Morella cerifera*
Myrica heterophylla	*Morella caroliniensis*
Myrica inodora	*Morella inodora*
Myriophyllum brasiliense	*Myriophyllum aquaticum*
Myrsine guianensis	*Myrsine floridana*
Nasturtium officinale	*Rorippa nasturtium-aquaticum*
Nasturtium sarmentosum	*Rorippa sarmentosa*
Nymphaea tuberosa	*Nymphaea odorata*
Nyssa sylvatica var. biflora	*Nyssa biflora*
Panicum clandestinum	*Dichanthelium clandestinum*
Panicum occidentale	*Dichanthelium acuminatum* var. *fasciculatum*
Peltiphyllum peltatum	*Darmera peltata*
Pentaphylloides floribunda	*Dauphora floribunda*
Petasites palmatus	*Petasites frigidus* var. *palmatus*
Phragmites communis	*Phragmites australis*
Pinckneya pubens	*Pinckneya bracteata*
Pluchea purpurascens	*Pluchea odorata* var. *odorata*
Poa alpigena	*Poa pratensis*
Polygonum minus	*Polygonum persicaria*
Polygonum opelousanum	*Polygonum hydropiperoides*
Potentilla anserina	*Argentina anserina*
Potentilla fruticosa	*Dauphora floribunda*
Potentilla palustris	*Comarum palustre*
Psoralea macrostachya	*Hoita macrostachya*
Psoralea orbicularis	*Hoita orbicularis*
Psychotria sulzneri	*Psychotria tenuifolia*
Pteretis pensylvanica	*Matteuccia struthiopteris*
Puccinellia airoides	*Puccinellia nuttalliana*
Quercus falcata var. pagodifolia	*Quercus pagoda*
Quercus nuttallii	*Quercus texana*
Ranunculus carolinianus	*Ranunculus hispidus var. nitidus*
Ranunculus septentrionalis	*Ranunculus hispidus var. nitidus*
Ranunculus subrigidus	*Ranunculus longirostris*
Rhexia glabella	*Rhexia alifanus*

Synonym (old name)	Currently Accepted Name
Rhododendron coryi	*Rhododendron viscosum*
Rhododendron serrulatum	*Rhododendron viscosum*
Rudbeckia amplexicaulis	*Dracopis amplexicaulis*
Rumex fascicularis	*Rumex verticillatus*
Rumex floridanus	*Rumex verticillatus*
Sagittaria falcata	*Sagittaria lancifolia*
Salix fluviatilis	*Salix melanopsis*
Salix interior	*Salix exigua*
Salix lasiandra	*Salix lucida* ssp. *lasiandra*
Salix longifolia	*Salix exigua*
Salix piperi	*Salix hookeriana*
Salix rigida	*Salix eriocephala*
Salix X subsericea	*Salix petiolaris*
Sambucus canadensis	*Sambucus nigra* ssp. *canadensis*
Sambucus melanocarpa	*Sambucus racemosa* var. *melanocarpa*
Scirpus acutus	*Schoenoplectus acutus var. acutus*
Scirpus americanus	*Schoenoplectus americanus*
Scirpus californicus	*Schoenoplectus californicus*
Scirpus fluviatilis	*Bolboschoenus fluviatilis*
Scirpus lacustris ssp. validus	*Schoenoplectus tabernaemontani*
Scirpus maritimus	*Schoenoplectus maritimus*
Scirpus pungens	*Schoenoplectus pungens var. pungens*
Scirpus robustus	*Schoenoplectus robustus*
Scirpus rubrotinctus	*Scirpus microcarpus*
Scirpus tabernaemontani	*Schoenoplectus tabernaemontani*
Scirpus torreyi	*Schoenoplectus torreyi*
Scirpus validus	*Schoenoplectus tabernaemontani*
Sesbania exaltata	*Sesbania herbacea*
Sesbania macrocarpa	*Sesbania herbacea*
Shinnersia rivularis	*Trichocoronis rivularis*
Sium floridanum	*Sium suave*
Smilacina stellata	*Maianthemum stellatum*
Smilacina trifolia	*Maianthemum trifolium*
Solidago confinis	*Solidago spectabilis* var. *confinis*
Solidago edisoniana	*Solidago latissimifolia*
Solidago elliottii	*Solidago latissimifolia*
Solidago mirabilis	*Solidago latissimifolia*
Solidago ohioensis	*Oligoneuron ohioense*
Solidago riddellii	*Oligoneuron riddellii*
Sparganium emersum	*Sparganium angustifolium*
Sphenostigma coelestinum	*Calydorea coelestina*
Spiraea latifolia	*Spiraea alba* var. *latifolia*
Stachys rigida	*Stachys bergii*
Tamarix tetrandra	*Tamarix parviflora*
Taxodium distichum var. nutans	*Taxodium ascendens*
Thelypteris thelypterioides	*Thelypteris noveboracensis*
Trilisa odoratissima	*Carphephorus odoratissimus*
Trilisa paniculata	*Carphephorus paniculatus*
Typha angustata	*Typha domingensis*

Synonym (old name)	Currently Accepted Name
Uniola latifolia	*Chasmanthium latifolium*
Utricularia vulgaris	*Utricularia macrorhiza*
Vaccinium microcarpos	*Vaccinium oxycoccos*
Vaccinium occidentale	*Vaccinium uliginosum*
Vallisneria spiralis	*Vallisneria americana*
Viburnum cassinoides	*Viburnum nudum* var. *cassinoides*
Viburnum nashii	*Viburnum obovatum*
Viburnum recognitum	*Viburnum dentatum var. lucidum*
Viburnum semitomentosum	*Viburnum dentatum var. dentatum*
Viburnum trilobum	*Viburnum opulus var. americanum*
Viola floridana	*Viola sororia*
Viola langloisii	*Viola affinis*
Viola pallens	*Viola macloskeyi ssp. pallens*
Viola pensylvanica	*Viola pubescens* var. *pubescens*
Wisteria macrostachya	*Wisteria frutescens*
Zosterella dubia	*Heteranthera dubia*

To Contents

United States
Department of
Agriculture

**Natural
Resources
Conservation
Service**

Wetland Restoration, Enhancement, and Management

Section II Ecological Monitoring

II.A Why monitor?

(Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001)

Purpose

- Distinguish between the processes of monitoring, management, and maintenance in wetland restoration and enhancement.
- Identify the benefits of monitoring and the key components of a wetland to be monitored.
- Describe the relationship between monitoring and maintenance to the success of a wetland restoration or enhancement activity.

Contents

Wetlands are valuable natural resources. They have the ability to provide a multitude of functions, many of which are valuable to us as individuals and to our society. From a plant and wildlife perspective, wetlands are equally valuable in that they provide living space, food, shelter, and other components of habitat for a suite of wetland dependent species. When wetlands are lost, so are these associated functions and values. When wetlands are restored and/or enhanced, the *potential* exists to replace lost functions and values. The extent to which the potential replacement of functions and values are realized depends, in a large part, on the effectiveness of monitoring and how well the physical and biological components of the wetland are maintained.

Planning is involved in recreating a wetland on the landscape. It begins with a visualization of the project's overall appearance and an understanding of the intended functions. Based upon the site's capability (i.e., water source, landscape position, plant propagule source) the next step is to determine the infrastructure, or physical components, necessary to produce the intended result. The positioning of berms and the designing of micro- and macrotopographic complexity are used to restore and enhance the wetland hydrology to meet the project criteria. These physical components are designed to allow the restored wetland to *operate* as planned and to set the stage for the *development* of the biological components, which create many of the wetland functions and values. Unlike the design, construction, and operation of the wetland infrastructure, the development of the biological components and the associated functions and values is a long-term process.

A contract may be let, the components may be installed, and the design completely implemented, but time is necessary for all the components to develop and operate together making a functional wetland ecosystem. Given a perfect world, the effect time has on the developing ecosystem is predictable (i.e., everything goes according to plan, is accomplished on schedule, and all within specifications). However, reality is much different in that the events that occur over time are not predictable. In short, the "mental model" of the wetland planned may not be the resultant wetland restored. At this point a restorationist must ask some serious questions.

- Does the developing wetland (that is not as planned) need to be reworked or is it still a valuable wetland even though it is not as planned?
- Does it still meet program objectives, landowner expectations, or planned functions and values?
- Is this wetland developing into a functioning wetland ecosystem despite that it has developed along a different trajectory than planned?
- Can you (or the landowner, program objectives) "live" with the differences?
- Is it necessary to rework the project?

Hopefully, one critical question that should be asked, given this scenario is, "How could I have detected this change in trajectory and what could I have done about it?" This last question can be answered in two words: monitoring and maintenance.

Definitions

Management is the application of ecological principles (generally) for increased production of wildlife species. With respect to wetland restoration and enhancement, it is planning and performing a regular set of activities that enables the site to fulfill its intended function; i.e., it is how the wetland is operated. This can include the manipulation of water levels to support specific wildlife species or to support specific species at specific times; controlling populations of beaver, nutria, or other nuisance wildlife species; or promoting the growth of specific plant species.

Monitoring is the long-term assessment of the condition of the wetland restoration and enhancement project. This includes inspection of installed **physical** components (berms, spillways, risers) and the **biological** components (vegetation and wildlife use). For programmatic purposes, monitoring may also include compliance checks for identified compatible uses and/or site restrictions.

Maintenance is performance of corrective action necessary to ensure proper operation of the installed components. It also includes manipulations necessary to "bring back" or "correct" the biological components of the system to keep the wetland in the planned successional trajectory, or lack there of. This would include activities, such as repair to berms or water control structures, control of invasive species, or removal of perennial vegetation.

Why is monitoring important?

- Provides a method for assessing a wetland's progression towards its planned objectives and the development of its planned functions.
- Provides documentation on the success in establishing the targeted vegetation, providing proper hydrology for the intended purposes, and patterns of wildlife use.
- Identifies failures or shortcomings that may threaten the project.
- Serves as a feedback mechanism to stimulate maintenance.
- Indicates need for changes to the management practices.
- Serves as a learning process. Future wetlands are restored and enhanced at a higher quality based upon knowledge of the successes and failures of earlier projects.

Components to be monitored

Not all wetlands are created equal. Regional differences, wetland types, project size, project goals, target wildlife, site conditions, contributing watershed, and program priorities are only a few factors that create this inequality. As such, it is unrealistic to identify a "one size fits all" monitoring protocol. However, evaluating categories of information in a monitoring exercise that have been modified to fit the project's geography, wetland type, functions, and objectives should be the approach. Hydrology, vegetation, wildlife, and time are the major components to be evaluated. The evaluation would also include any structures or physical aspects installed to support or enhance these components. Through these four sources of information, the functions and objectives can be evaluated and whether they are developing at an appropriate rate can be determined.

Frequency of monitoring depends upon the complexity of the project, the intended outcome, and the potential for problems to develop. During the early development stages of the wetland, monitoring should occur frequently; i.e., year 1 and year 3. This allows assessment as to whether the wetland is functioning as designed and developing as planned. After these initial stages, monitoring frequencies may decrease to once every 5 to 6 years. In determining a monitoring frequency, the project and the potential for difficulties to arise must be considered. Will the area be flooded with water containing seed of invasive species? Are nutria, beaver, or other nuisance wildlife a high risk? Is the site adjacent to a large watercourse with a history of forceful flooding that can impact installed structures? These, as well as other factors, will subject the restoration and enhancement site to greater risk of failure. It is worth considering an increased monitoring frequency if these or other high-risk scenarios are real possibilities.

Ecological Monitoring

Elements to Monitor

Hydrology
- Hydrology for each hydrology zone (use Cowardin system modifiers)
- Percent of wetland at different hydrology zones (open water, semi-permanent, seasonal)

Vegetation
- Percent cover of the three dominant species
- Density of targeted species with planting objectives
- Density and type (annual, perennial, woody) of targeted species for planned wildlife objectives
- Noxious and invasive species

Wildlife
- Overall wildlife use by targeted species
- Threatened and endangered species use (especially for those species identified during the ranking process)
- Habitat models for specific species
- Nuisance species

Frequency
- Evaluate: year 1
- Monitor years: 3, 6, 12, 18, 24, (year + 6)

II.E Monitoring hydrology/ structures

(Paul Rodrigue, NRCS Wetland Science Institute, Oxford, Mississippi, December 2001)

Purpose

This paper provides basic guidance on establishing a monitoring program for hydrology and related structural components on wetland restoration or enhancement projects.

Contents

Monitoring hydrology

Hydrology should be monitored to ensure that the planned hydroperiod has been achieved. By monitoring hydrology, failure in any part of the wetland restoration can be linked or dissociated from hydrology based upon monitored hydrology data rather than pure conjecture.

Monitoring may be simple and inexpensive (staff gage) or intensive and expensive (continuous monitoring wells) depending upon the monitoring needs.

Photo points—Fixed photo points (a WRP requirement) can provide visual documentation of hydrology as long as the photo points are representative of the sites of interest and the timing of the pictures coincides with the hydroperiod of consequence. The addition of a fixed staff gage at the photo point can assist in measuring inundation.

Observation wells—If soil saturation (water table) is of primary concern, simple PVC observation wells can be installed and water table measurements made at appropriate times to coincide with the period of interest. Establishing a photo point at well locations is also recommended. Additional information on observation well installation and use is available at **http://www.wes.army.mil/el/wrtc/wrp/tnotes/hyia3-1.pdf.**

Recording wells can be installed at remote sites or sites needing or warranting a continuous hydrology record. Recording wells need to be downloaded only once every 6 months. The expense (ex. $800) per well needs to be justified by monitoring needs and objectives.

Monitoring structures

Structures should be monitored for proper functioning and possible degradation of integrity.

Dikes—Dikes should be monitored for proper cover, erosion (rilling, overtopping, wave action), animal damage (burrowing animals), soil cracking, and other such concerns. Any of these problems could lead to physical failure of the dike.

Water control structures—Water control structures should be monitored for proper functioning condition, integrity, rust, beaver activity, and other related problems. The operational status should also be monitored (are stoplogs installed/removed in a timely fashion in accordance with the compatible use permit). Compliance with the management plan and compatible use permits should be monitored and evaluated.

United States
Department of
Agriculture

**Natural
Resources
Conservation
Service**

Wetland Restoration, Enhancement, and Management

Section III Management

III.D.6 Managing Wildlife Groups—Reptiles and Amphibians

(Laura Mazanti, NRCS Wetland Science Institute, Laurel, Maryland, December 2001)

Purpose

The purpose of this paper is to provide information on factors affecting the colonization and breeding success of amphibians and reptiles in wetland restoration and enhancement projects. Specific management actions and physical features that may improve the habitat quality of restored and enhanced wetlands by amphibians and reptiles are identified.

Contents

External factors that affect the colonization and breeding success of amphibians and reptiles in restored and enhanced wetlands are roads, road culverts and ditches, terrestrial buffers, and connectivity to wetland and upland habitat.

Roads

Amphibians and reptiles are negatively impacted by roads that are close to or intersect their aquatic and terrestrial habitats. Species abundance and diversity can strongly decrease when a highway is located within 2 miles of a population by acting as a barrier to dispersal and breeding migrations (Vos and Chardon 1998, deMaynadier and Hunter 2000). Findlay and Houlahan (1997) found that species richness of amphibians and reptiles decreased with increasing density of roads located up to 1.25 miles from a wetland. They also found that a 9 square foot per acre increase in hard surface road density might cause a 19 percent decline in frog species richness in nearby wetlands and pools. The exchange of genetic material within reptile and amphibian populations is restricted when roads separate habitats by increasing inbreeding among closely related individuals. This may result in low genetic heterozygosity, which in turn renders populations more susceptible to demographic stochasticity and catastrophic events that threaten their long-term viability (Reh and Seitz 1990).

Other attributes of roads also make them hazardous to amphibian and reptiles. Paved and dirt roads can alter natural microclimatic conditions up to 100 feet from the shoulder, including temperature, humidity, and evaporation (Mader 1984). Because amphibians must maintain body moisture content to prevent desiccation, a hot or dry road surface can be effectively impassable. Natural movement between aquatic and terrestrial habitats is also disturbed by the noise, artificial night-light, exhaust fumes, and sand and salt runoff associated with roadways (Mader 1984). Although frogs are often observed in roadside ditches and right-of-ways, unstable physical conditions, such as periodic mowing and herbicide spraying, degrade the habitat quality of the area and most likely prevent long-term viability of a population. Roads also cause direct mortality as well as indirect population level adverse effects. Reh and Seitz (1990) found that traffic related mortality from crossing roads could decrease species abundance in adjacent habitats by 50 to 70 percent. They also found that if traffic density on a roadway is greater than 26 cars passing per hour, 50 to 100 percent of migrating amphibians might be killed when attempting to cross it.

Road culverts and ditches

Road culverts positively affect the success of amphibians and reptiles crossing a roadway, permitting safe mobility among natural habitats intersected by paved and dirt roads. Yanes et al. (1995) found that lizards and snakes readily accept culverts to cross under roadways. In addition, culverts were used preferentially by amphibians to cross roadways on rainy, spring nights to the extent that no amphibians were observed crossing the road surface when a culvert was available (Yanes et al.1995). In terms of size, amphibians and reptiles can generally use small to medium culverts (16- to 24-inch diameter), but installing larger culverts (>24-inch diameter) provides access for a wider range of species. Box culverts with the bottom open to the natural ground substrate may be more conducive to passage than culverts with a concrete or metal bottom.

Vegetative diversity and density in the right-of-way leading up to the culvert can affect the utility of the culvert to amphibians and reptiles. Typical highway shoulder vegetation is characterized by grass monocultures. This should be supplanted in the area around the mouth of the culvert with native herbaceous and shrub plant species to enhance microclimatic conditions and provide cover. Ditches that convey surface water to culverts often increase movement of herptiles between habitats (Reh and Seitz 1990), especially those with complex vegetative structure and species diversity as compared to those that are regularly mowed, sprayed, or denuded of vegetation. It has been observed that amphibian populations in habitats connected by vegetated ditches have higher genetic diversity and lower genetic isolation than habitats not connected by ditches.

Physical features often associated with culverts and roadways, such as debris pits, earthen berms, and riprap armor, can detract from the amphibian and reptile use of culverts (Yanes et al. 1995). Yanes et al. found that debris pits, when constructed at the mouth of culverts, restrict their entrance into the culvert, and should be eliminated from the design or modified using ramps. Earthen berms and riprap armor also impede access to culverts, acting as barriers to migration. Increasing the abundance and diversity of vegetative species on berms, reducing the size of berms, and eliminating riprap improve access for herptiles. If rock armor is necessary, use as little as possible (one layer only, not mounds of riprap) and add soil and herbaceous vegetation within the rock crevices.

Terrestrial buffer

Many frogs and toads have distinct aquatic and terrestrial life stages; e.g., breeding in wetland pools in the spring and retreating to underground burrows during the winter. Turtles also use multiple habitat types, with some species, such as Florida cooters (*Chrysemys floridana*) and yellow bellied sliders (*Chrysemys scripta*), overwintering in ponds and wetlands, and others, such as mud turtles, hibernating in terrestrial burrows (Burke and Gibbons 1995). Studies have shown that during nonbreeding times of year, up to 95 percent of a salamander population may be in a 550-foot-wide forested area adjacent to wetland pools (Semlitsch 1998). Dodd and Cade (1998) found that such a buffer must have a directional as well as distance component that is tailored to the specific life history requirements of the amphibians using the wetland. Protecting this forested habitat as a buffer is as critical to the survival of amphibians and reptiles as is protecting the wetland. Without it, the water quality of the wetland would be degraded and essential upland foraging, hibernation, and migration habitat would be lost. Similarly, Burke and Gibbons (1995) found that a terrestrial buffer of at least 900 feet beyond the wetland boundary would be required to ensure that the nesting, hibernating, foraging, and other life history attributes of mud turtles, Florida cooters, and slider turtles would be met. Studies have shown that amphibian species richness decreases when forest cover in buffer areas and migration corridors declines. For example, a 20 percent loss of forest cover within 1.25 miles of a wetland pool can result in a 17 percent decline in amphibian species richness, which is equal to the amphibian loss that would result from the loss of 50 percent of the wetland itself (Laan and Verboom 1990).

Connectivity to wetland and upland habitat

Many amphibians and reptiles emigrate from natal wetlands to new wetland pools when competition for space and food in their home pond becomes too great or physical conditions deteriorate. They often travel hundreds of meters overland (Pechmann et al. 1989). For example, Gibbons et al. (1983) found that prolonged drought conditions caused two species of aquatic turtles (*Pseudemys scripta* and *P. floridana*) to emigrate 250 miles from their home wetland to a beaver pond. Migratory routes are disrupted by intensively managed agricultural fields that fragment and isolate formerly large, continuous natural habitats, which in turn increases the likelihood of higher mortality of amphibians during migration and lower survival of the local population (Vos and Stumpel 1995). Cultivated land presents a barrier to migration as the temperature, moisture content and cover on the ground surface are inhospitable to amphibians and reptiles, and farm equipment and pesticide applications can be directly detrimental to their survival. Amphibians and reptiles may rely on environmental or physical cues to orient migration to new habitats when their home habitat quality deteriorates (Yeomans 1995). While the exact cueing mechanisms remain unknown, alteration of natural environmental conditions between aquatic habitats may inhibit the ability of turtles and frogs to locate nearby waterbodies that are unfamiliar and out of their sight.

Fragmentation of natural habitat alters the edge to interior habitat size ratio, giving non-native and invasive species of plants and animals the opportunity to colonize remaining natural areas, which further threatens the survival of native herptiles (deMaynadier and Hunter 1998). As species composition changes with decreasing habitat area, natural competitive outcomes between the remaining native species may be altered such that the viability of the community is threatened. Decreasing species abundance and distribution in a habitat might also decrease the stability and self-sustaining nature of the population and community. Alternatively, restoring or enhancing multiple wetland pools within a complex of existing natural upland and wetland habitats would maximize the potential amphibian colonization of the new wetland and improve the long-term viability of amphibian populations.

Recommendations

Wetland restoration actions to increase colonization and breeding success by amphibians and reptiles

Vernal pools and other temporary wetlands— Vernal pools and other temporary wetlands with standing water have hydrologic regimes that are characterized by periodic inundation and drying, including northeastern vernal pools, midwestern prairie potholes, southwestern playas and flood plain wetlands. These wetlands provide habitat for winter and spring breeding amphibians and reptiles that cannot survive in permanent pools.

Recommendations to enhance vernal pool and temporary wetlands are as follows:

- Incorporate macrotopographic features, such as depressions and pools, within the broader wetland area.
- Place large logs, rocks, and debris piles within pools and in the shallows to provide basking and loafing habitat for turtles and snakes. Logs in deeper portions of the pool may need to be anchored in place. In terrestrial areas, construct snake mounds consisting of stumps, rocks, logs, brush, and sand/soil that are partially buried and are 4 to 6 feet high.
- Vegetate the littoral zone and pools with submerged and herbaceous plants to provide structure for egg mass attachment and refugia for larvae and juvenile herptiles.
- Make pool size and depth consistent with that of naturally occurring pools in the area because local species, from which recruits to the restored wetlands are drawn, are adapted to particular hydric regimes. Snodgrass et al. (2000) found that species composition in wetlands changes along with increasing or decreasing hydroperiod; short hydroperiod wetlands (which tend to be small) support unique amphibian species found only in such wetlands. The insect prey base is connected to the duration of ponded water, which in turn influences the types of invertebrate and vertebrate predators that use a wetland. For example, wetlands with long hydroperiods that dry for only a few months have somewhat slow growing, inconspicuous feeding predator populations, while highly ephemeral wetlands support rapid development and conspicuous feeding insect predators to improve the chances that an individual will reach metamorphosis before the wetland dries up.
- Exclude fish and large frog species (e.g., bullfrogs) that prey upon the eggs, larvae, and juveniles of spring breeders. The presence of fish and other vertebrate predators in a wetland with a permanent pool eliminates all but the most cryptic and slow developing species (Snodgrass et al. 2000).
- Restore or create multiple pools or depressions within a few hundred feet of one another to provide dispersal opportunities for juveniles and migrating adults. Multiple wetland habitats also offer refugia to animals displaced from their home wetland by natural events, such as drought. They provide alternative habitat patches from and to which immigration and emigration can occur when conditions in a home pool deteriorate, thereby sustaining the population long-term.
- Eliminate or reduce sources of sediment, nutrients, pesticides or salts that may enter the wetland via surface water runoff from roads or fields.
- Create, restore, and protect shrub and forested habitat adjacent to the new wetlands, existing wetlands, and between crop fields (deMaynadier & Hunter 1999).
- Provide large, continuous blocks of terrestrial, forested habitat within several hundred feet of the wetland or in a 500-foot buffer zone around the wetland. This is necessary to provide wintering and migratory habitat.
- Avoid creating or maintaining narrow, linear corridors (< 100 ft wide) as these may be insufficient for herptile movement due to increased negative edge effects that allow invasive or predaceous species to the interior of the corridor.

- Restore or locate wetlands that are away from dirt or paved roads (hundreds to thousands of feet away) to minimize road-related adverse effects.
- Install culvert(s) under the roadway (when proximity to a road is unavoidable) to connect wetland and terrestrial habitat, including naturally vegetated, lightly or unmanaged rights-of-way that lead amphibians and reptiles to the culvert opening.

Wetlands with permanent standing pools— Wetlands in this category are those with hydrologic regimes characterized by permanent standing pools. These wetlands support amphibians and reptiles that use aquatic habitats year-round (e.g., aquatic turtles, leopard frogs, and water snakes) and provide refugia during periods of drought or drawdown.

Recommendations to enhance wetlands with permanent standing pools:

- Include gradually sloped, vegetated littoral zones and mudflats to provide loafing, foraging, and calling habitat.
- Place large logs in the shallows and anchor them into deeper water to provide basking habitat for turtles. Create snake mounds made of stumps, woody debris, and rocks in terrestrial areas for loafing, foraging, and hibernacula for snakes and other reptiles.
- Plant or augment standing pool with native submerged aquatic vegetation.
- Stock the permanent pool with fish or physically connect it to existing pools that support fish.
- Define pool dimensions consistent with those naturally occurring on the landscape or those consistent with nearby created wetlands that do support amphibian and reptile populations.
- Avoid large, earthen or riprap berms in the wetland design. If unavoidable, berms should be vegetated with native herbaceous, shrub and tree species and incorporated in the larger terrestrial buffer area surrounding the wetland.
- Vegetate the terrestrial buffer area around the wetland with herbaceous and shrub species, and create a transition into a larger forested buffer that together extend at least 500 feet from the wetland edge to provide wintering and migratory habitat.
- Avoid narrow, linear corridors (< 100 ft wide) that may be insufficient for herptile movement due to increased negative edge effects that allow invasive or predaceous species into the interior of the corridor.
- Restore or locate wetlands that are distant from dirt or paved roads (hundreds to thousands of feet away) to minimize road-related adverse effects.
- Install culvert(s) under the roadway (when proximity to a road is unavoidable) to connect wetland and terrestrial habitat, including naturally vegetated, lightly or unmanaged rights-of-way that lead amphibians and reptiles to the culvert opening.

References

Burke, V.J., and J.W. Gibbons. 1995. Terrestrial buffer zones and wetland conservation: a case study of freshwater turtles in a Carolina bay. Conservation Biology 9(6):1365–1369.

deMaynadier, P.G., and M.L. Hunter, Jr. 1998. Effects of silvicultural edges on the distribution and abundance of amphibians in Maine. Conservation Biology 12:340–352.

deMaynadier, P.G., and M.L. Hunter, Jr. 1999. Forest canopy closure and dispersal by pool-breeding amphibians in Maine. J. Wildlife Management 63:441–450.

deMaynadier, P.G., and M.L. Hunter, Jr. 2000. Road effects on amphibian movements in a forested landscape. Natural Areas Journal 20 (1):56–65.

Dodd, C.K., and B.S. Cade. 1998. Movement patterns and the conservation of amphibians breeding in small, temporary wetlands. Conservation Biology 12(2):331–339.

Findlay, C.S. 1997. Anthropogenic correlates of species richness in southeastern Ontario wetlands. Conservation Biology 11(4):1000–1009.

Gibbons, J.W., J.L. Greene, and J.D. Congdon. 1983. Drought-related responses of aquatic turtle populations. J. Herpetology 17(3):242–246.

Laan, R., and B. Verboom. 1990. Effects of pool size and isolation on amphibian communities. Biological Conservation 54:251-262.

Mader, H.J. 1984. Animal habitat isolation by roads and agricultural fields. Biological Conservation 29:81–96.

Pechmann, D.E. Scott, J.W. Gibbons, and R.D. Semlitsch. 1989. Influence of wetland hydroperiod on diversity and abundance of metamorphosis juvenile amphibians. Wetlands Ecology and Management 1(1):3–11.

Reh, W., and A. Seitz. 1990. The influence of land use on the genetic structure of populations of the common frog *Rana temporaria*. Biological Conservation 54:239–249.

Semlitsch, R.D. 1998. Biological delineation of terrestrial buffer zones for pond-breeding salamanders. Conservation Biology 12(5):1113–1119.

Snodgrass, J.W., M.J. Kmoroski, A.L. Bryan, Jr., and J. Burger. 2000. Relationships among isolated wetland size, hydroperiod, and amphibian species richness: implications for wetland regulations. Conservation Biology 14(2):414–419.

Vos, C.C., and H.P. Stumpel. 1995. Comparison of habitat-isolation parameters in relation to fragmented distribution patterns in the tree frog (*Hyla arborea*). Landscape Ecology 11(4):203–214.

Vos, C.C., and J.P. Chardon. 1998. Effects of habitat fragmentation and road density on the distribution pattern of the moor frog *Rana arvalis*. J. Applied Ecology 35:44–56.

Yanes, M., J.M. Velasco, and F. Suarez. 1995. Permeability of roads and railways to vertebrates: the importance of culverts. Biological Conservation 71:217–222.

Yeomans, S.R. 1995. Water-finding in adult turtles: random search or oriented behaviour? Animal Behaviour 49:977–987.

III.E.3 Managing for potential problem species (beavers and muskrats)

(Paul Rodrigue, NRCS Wetland Science Institute, Oxford, Mississippi, December 2001)

Purpose

This paper provides information on the implications of beaver and muskrat activity on wetland restoration and enhancement projects, particularly on structural measures. Internet sites where more detailed and specific information can be found are provided.

Content

Beaver, muskrat, and other forms of wildlife may become biological nuisances on a wetland site, rather than a benefit, if their activities compromise the integrity and operation of structures, such as dikes, embankments, and water control structures.

Beaver and other animals can develop natural and diverse hydrology (beaver ponds) and are often desired on a wetland restoration site. However, their activity can negatively affect the hydroperiod (backing water off a site) as well as vegetation (destruction of planted woody species). Hunting and trapping may be required to maintain a balanced population, especially if natural predators are absent.

Where possible, excavation as a source of fill for dikes should be away from the dike. This prevents the establishment of permanent water against the dike and the inherent possibility of providing habitat for burrowing rodents.

Beaver

Managing beaver pond water levels

Beaver ponds can be drawn down by the use of such means as the Clemson Beaver Device. Installed through the beaver dam, it allows manipulation of the water level in the beaver pond while preventing further control by the beaver.

Tree protection

Using beaver fences to protect trees has had some success. A 6-foot or so roll of wire is used, and it is posted up 3 feet high with 3 feet of wire flat on the ground. The beaver/nutria walk up to the fence and try to dig under it, but cannot dig through the fencing. The horizontal part of the fencing is staked to the ground.

Beaver damage

Beaver prevention devices are often promoted as protection from beavers around water control structures. Success is inconsistent and difficult to judge. A study was done by the Mississippi NRCS in 1990 on evaluation of trapping for beaver control on floodwater retarding structures. Previous mechanical and structural modifications to reduce or eliminate beaver impairment to principal spillways failed in 75 percent of cases, primarily due to lack of maintenance. The study found that hunting and trapping to control beaver populations on these lakes was an effective control of beaver activity. An initial effort to reduce populations would need followup every 2 to 3 years to maintain low population densities.

Open flow structures, as opposed to conduit structures, present less risk of beaver manipulation and represent easier maintenance if beaver become active. If beavers are in the area, the wetland restoration planner should anticipate beaver activity and locate flowing water features where beaver activity will have the least negative impact while incorporating the beneficial impact of beaver activity.

Muskrats

Muskrats can cause considerable damage to dikes and embankments on water impoundments. Excavated water features, as opposed to embankment features, can reduce the structural impacts of muskrats. The lower cost of embankment impoundments must be weighed in light of continual embankment operation and maintenance requirements.

Wider levees can minimize the impacts of burrowing animals, such as muskrat, beaver, and nutria. Wire mesh can be incorporated into the face of embankments in extreme cases, but becomes impractical in large restoration sites. Hunting and trapping remain an effective component of a long-term operation and maintenance program.

Additional resource information

http://www.uaex.edu/aquaculture2/FSA/FSA9068.htm, Flood Water Management with a Beaver Pond Leveler, Dr. Bert Bivings, Extension Wildlife Specialist, Phil Tacker, Extension Agricultural Engineer

http://www.ces.ncsu.edu/nreos/forest/steward/www23.html, Managing Beaver Ponds

http://www.ces.ncsu.edu/nreos/wild/wildlife/beavers.html, Wildlife Damage Management: Beavers, AG–472–4, North Carolina Cooperative Extension Service

http://www.beaversww.org/, Beavers, Wetlands and Wildlife

III.G Decisionmaking, design, and management of greentree reservoirs

(Sammy King, University of Tennessee Department of Forestry, and Leigh Fredrickson, University of Missouri Gaylord Laboratory, December 2001)

Purpose

This paper contains information for resource managers and other interested parties on greentree reservoir (GTR) management projects. It includes a step-by-step guide on GTRs from the beginning processes of deciding whether a GTR is appropriate for the site and restoration/enhancement project focus through GTR design and long-term management processes. An ecological monitoring format and protocol are included to aid identifying ecosystem stresses and addressed in management plans.

Contents

(At the time of printing, this paper was unavailable for inclusion.)

III.I Managing native grass stands

(John Dickerson, NRCS Syracuse, New York, December 2001)

Purpose

To provide information on post-planting and post-establishment management for native warm-season and cool-season grasses used in buffers and upland inclusions within wetlands. Specific information on the management tools including clipping, burning, herbicide use, and grazing are included.

Contents

Most wetlands are surrounded by associated upland areas that can be managed to enhance the value of the wetland. Native warm-season grasses are particularly valuable when associated with wetlands. While some wetlands have naturally occurring grasslands surrounding them, others can be planted to native grasses as site enhancement. In both cases, long-term management is critical to maintain the grass cover. Most grasslands shift to other cover types without management; most often to brush or woodland. This section provides guidance for managing warm-season grasslands whether they are planted or naturally occurring. The information presented here is largely extracted from *Vegetating with Native Grasses in Northeastern North America*, a joint publication of Ducks Unlimited Canada and the USDA Natural Resources Conservation Service, November 1997.

Post-planting weed control in establishing native grass stands

Some references to specific herbicides and rates of application in this section are based on preliminary research and field experience. Unless stated otherwise, they should NOT be construed as recommendations for herbicide use under differing conditions and label recommendations. For specific recommendations for local conditions, refer to State or provincial weed control publications, herbicide label information, and experienced local personnel.

Planting year

Prompt attention to post-plant weed control is required on all sites during the establishment year. Weed control options and strategies vary depending on the type of planting (cool-season, warm-season, or mixed grasses), the weed species present, whether forbs or legumes have been included, and whether the stand will be harvested for forage.

Weed control in native warm-season grasses is particularly important to allow the slowly developing seedling to receive direct sunlight. Native warm-season grass seed germinates slowly and preferentially uses the endosperm energy to promote root growth rather than leaf growth. This drought-tolerance strategy is a problem under moist conditions because weeds can produce dense shade over the first leaves at a time when they must capture sunlight to start photosynthesis and power the continued development of the seedling. Most cool-season grasses have a different strategy in which they preferentially produce rapid leaf growth and compete strongly for sunlight.

Four basic methods can be used to control weeds in developing grass stands: clipping, herbicides, grazing, and fire. Of these, only the first two are normally used during the planting year.

Clipping is the simplest method of assisting establishment of new grass stands, especially warm-season grasses. The goal is to reduce the shade pressure that the weeds are exerting and to keep the weeds from producing seeds. The best equipment to use for this method is a sickle bar mower that can operate horizontally within an elevation range of 6 to 12 inches (15–30 cm). Sickle bars are preferred over rotary mowers because they cut and drop each stem individually. Weeds are spread evenly over the entire swath, unlike the bunching that tends to happen with a rotary mower. However, clipping with a rotary mower is acceptable should a sickle bar be unavailable. **If possible, clipping should be delayed until ground-nesting birds have completed incubation, but this is often too late for warm-season grass seedlings.** Most sites with warm-season grasses require at least three clippings during the establishment year, and the early mowing(s) are the most critical to seedling success.

The height of cut is not critical if clipping cool-season grasses. Cool-season grasses generally have good seedling vigor, and the growing point (meristem) is near the soil surface where it is not likely to be cut off. It is generally acceptable to cut off some of the grass leaf with the weeds. Leaving two-thirds or more of the leaf length uncut is a good policy. Plants need leaf surface to capture light and generate food for growth, so cutting off half or more of the leaf can retard growth.

The weaker seedling vigor of the plants and an elevated growing point must be considered when first year, warm-season grass stands are clipped. With these grasses, only the leaf tips should be cut to avoid removing the meristem. Clipping either grass type can promote "stooling out" of the plant, which is the stimulation of basal buds to produce more stems and leaves.

Herbicides, used correctly, can effectively control many weeds in a timely and cost-effective manner. As with other weed control methods, the development of a herbicide use strategy should begin with a field inspection 4 to 6 weeks post-planting to identify the weed species present. The proximity of the grass stand to the wetland impacts the ability to use herbicides or may affect the formulation that can be used.

In the past, few herbicides were specifically tested for their efficacy on native plant materials. That situation has begun to change in recent years. Some of the new crop protection products carry registered uses for native plant species, either on their original labels or on supplemental labels that were issued soon after the products were released. In the United States, for example, some members of the imidazolinone family of herbicides carry such labeled uses. Masters et al. (1996) reported on a comparison of three of those products—Arsenal™, Plateau™, and Pursuit™—versus atrazine as part of an integrated weed management strategy to establish or restore warm-season grasses in the Great Plains.

Each of those herbicides control or suppress a range of annual, grassy and broadleaf weeds as well as some broadleaf and cool-season grass perennials, but they do differ in their effects on specific warm-season species. Plateau™, for example, is labeled for use on big bluestem, little bluestem, indiangrass, sideoats grama, blue grama, and buffalograss, while the Pursuit™ label covers big bluestem, little bluestem, and switchgrass.

The U.S. label for Plateau™ also contains tolerance information for a range of seedling and established forbs and legumes, including such species as black-eyed susan, yellow coneflower, and partridge pea. As a result, it may be a useful weed control product in plantings that include forbs and legumes or on degraded sites where restoration work is being undertaken. As is the case with all herbicides, the reader is urged to consult the product label for complete, specific information.

Even after a diligent weed-control program in the preplanting year(s), additional control measures are often required in the establishment year. Figure III.I–1 identifies considerations and strategies for weed control during the planting year. Annual broadleaf weeds are fairly easy to control in warm- or cool-season grass stands by using chemicals like 2,4-D or Banvel™. If annual grassy weeds are also present, other products or tank mixes of products may control them both in one application. Tables III.I–1 and III.I–2 outline a number of herbicide treatments that might be considered for both pre-seeding and post-emergent weed control during the planting year.

In special problem situations, such as cool-season grasses invading a warm-season stand, the herbicide options become more limited. In some situations, it is possible to use such products as atrazine or Plateau™. It may also be possible to apply Roundup™ to growing cool-season grasses while the warm-season grasses are dormant in early spring or late fall. If the warm-season grasses are dormant to the soil (green does not show) and the cool-season plants are actively growing, this treatment can provide some control.

Wick applicators can be used to apply Roundup™° to susceptible broadleaf weeds and grasses if they have grown above the desired plants, but this method is not effective if the weed population is high. Unless the wick method can be used, cool-season grass weeds in a stand of native cool-season grasses are essentially untouchable with herbicides.

Grazing can be used to advantage, but has several potential pit-falls. It must be carefully controlled or the stand can be destroyed by overgrazing, hoof damage,

Figure III.I–1 Planting year weed control

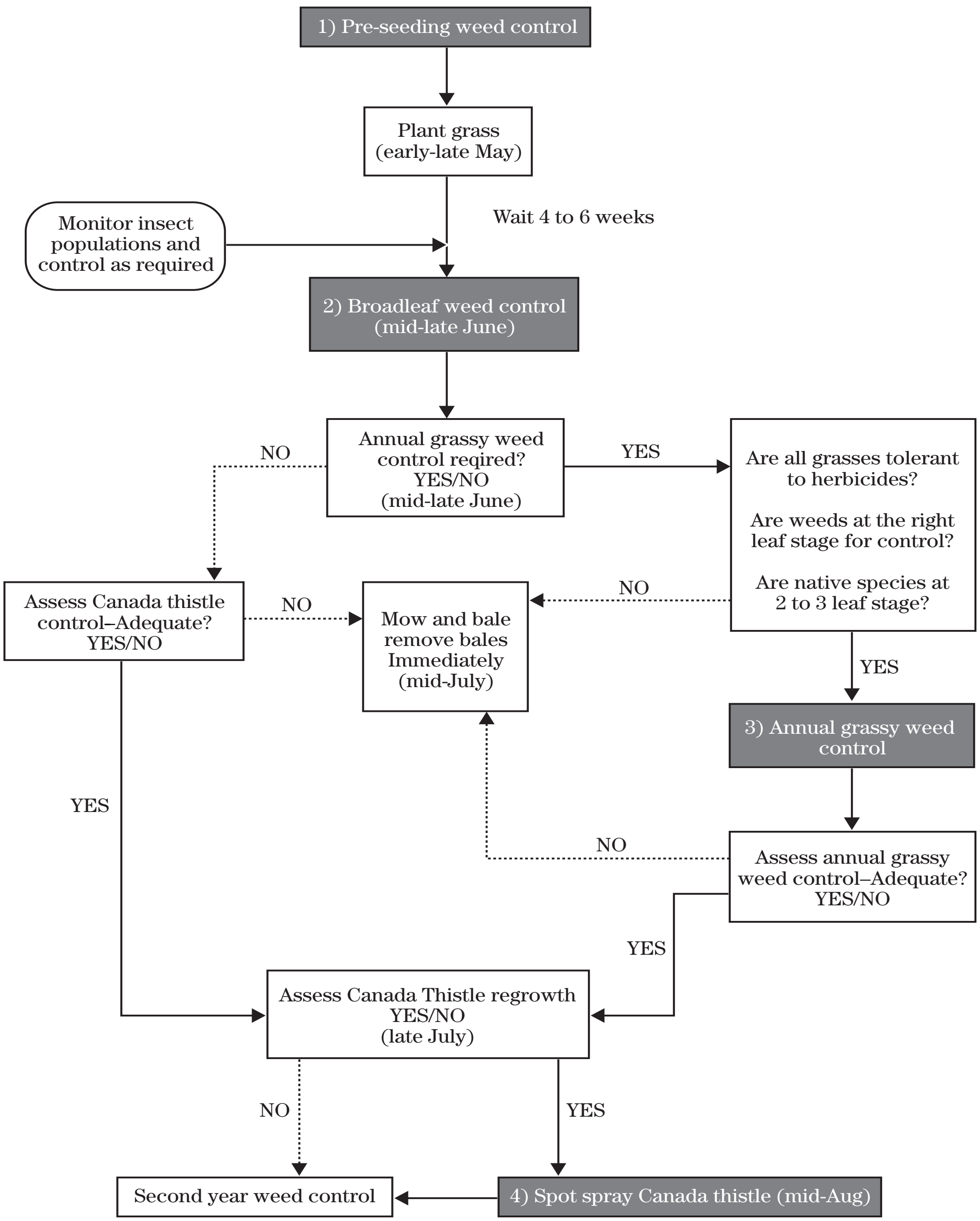


Table III.I–1 Planting year pre-seeding weed control *

Weed	Date	Herbicide	Rate (L/ac)	Chemical name	Active ingred. (g/L)	Active ingred. (g/ac)	Leaf stage crop	Leaf stage weed
Annual quackgrass weeds	various	Roundup™ Roundup™ + Pardner™	1.0 1.0 + 0.5	glyphosate glyphosate bromoxynil	356 356 280	356 356 140	n/a	< 6 inches
Annual broadleaf weeds	various	2,4-D amine	0.45	2,4-D	500	225	n/a	2 to 4

* Application rates are based on Canadian experience. For appropriate recommendations for U.S. conditions, contact your State Cooperative Extension Service.

Table III.I–2 Planting year post-emergent weed control *

Weed	Date	Herbicide	Rate (L/ac)	Chemical name	Active ingred. (g/L)	Active ingred. (g/ac)	Leaf stage crop	Leaf stage weed
Annual broadleaf weeds	mid – late June	Banvel™ + 2,4-D amine	0.2 + 0.4-0.6	dicamba 2,4-D	480 500	96 200–300	2 to 4	• 2 to 5 • Wild buck-wheat to 3 inches
		Target™	0.61	MCPA Mecoprop Dicamba	275 62.5 62.5	167 38 38	2 to 4	• 2 to 3
Canada thistle, annual broadleaf weeds	mid – late June	Lontrel™ + MCPA ester	0.17–0.34+ 500	clopyralid MCPA	360 500	61–122 170–225	2 to 4	• Majority of Canada thistle are 4 inches before bud stage, • Annual weeds—2–4 leaf
Annual grassy weeds	mid – late June	Hoe-grass™ ** 284	1.0 – 1.4	diclofop-methyl	284	284–398	2 to 3	1 to 4
Canada thistle	mid Aug	Lontrel™ + MCPA ester	0.11-0.34 + 0.34–0.45	clopyralid MCPA	360 500	40–122 170–225		• Fall regrowth (spot treat-ment)

* Application rates are based on Canadian experience. For appropriate recommendations for U.S. conditions, contact your State Cooperative Extension Service.

** Do not apply Hoe-grass 284 to plantings that contain warm-season grasses other than big bluestem and switchgrass.

or uprooting the seedlings as the animals bite and tear off the forage. In the second year and beyond, risk from hoof damage and the dislodging of plants are greatly reduced. Grazing can be used to reduce weed pressure early in the season where cool-season grasses are invading a warm-season grass stand. Livestock are attracted to the tender new growth of the cool-season grasses before the warm-season grasses begin to grow. Intense grazing at this time can weaken the cool-season grasses significantly. The animals' feed intake may need to be supplemented as grass growth slows. They should be removed as soon as the warm-season grass starts to grow. Sheep or goats can be used to selectively graze broadleaf weeds early in the season. If a planting is grazed, it is sometimes useful to build an enclosure to leave a small plot ungrazed so one can monitor plant growth. Grazing early affects the short-term value of the stand for nesting habitat.

Fire is an excellent tool to use to stress weeds in a warm-season grass stand, but the fire must be carefully controlled. Smoke in the wrong place is a hazard to road traffic and airplanes, and bothersome to neighbors. Local regulations must be met. Enlisting the aid of fire control experts, perhaps to use a controlled burn as a training exercise, is a good idea. Standing warm-season grass residue burns hot and fast. Slow backfires are not desirable because they can create a hotter soil temperature than fast moving fire does, and thereby damage the crowns of the plants. **If local authorities insist that a backfire must be used through the entire stand, abandon the plans to burn.** Generally, controlled burns should be spring events that are timed to take advantage of dry fuel and winds between 5 and 10 miles per hour from a favorable direction. The warm-season grasses should have 1 to 3 inches of new growth. After the burn, the black ash absorbs solar radiation, warms the soil rapidly, and in turn causes more rapid grass growth. Warm-season grasses gain advantage at the same time the weeds and cool-season grasses are set back. Fire is not a useful tool for managing cool-season grass stands though they can sometimes carry a fire in the spring.

Cool-season grasses can be used as an effective firebreak around warm-season stands, but they will carry a fire if not raked free of dead material and thatch. If in doubt, wetting the firebreak with water immediately before a burn is good insurance. Before providing assistance on a burn or burning recommendations, consult the NRCS Burn Policy in the General Manual. Before any use of fire is attempted, a plan must be in effect, necessary permits arranged, the right equipment and sufficient personnel to control the fire must be on hand, effective preparation of firebreaks must be achieved, and the weather and timing must be favorable.

Post-establishment management

Stand evaluation

Cool-season grass turf and lawns require high seedling density to create smooth walking surfaces and crowd out weeds under close, frequent mowings. Nearly everyone has had some experience with planting lawns, so the expectations and measures of success developed for that process tend to be carried over to other grass plantings, regardless of plant type or planting objective. This carryover can get in the way of objective stand evaluation and cause needless reseeding and repetitive effort with warm-season grasses. For turf establishment, thick stands of seedlings (one per square inch) are desired; for erosion control, forage, or wildlife cover managers can be successful with stands that are initially much less dense.

Wildlife, erosion control, and forage plantings need to grow much taller than turf plantings, so individual plant vigor is important. The relationship between individual plant size and rooting depth must also be considered. Bigger plants root more deeply and are, therefore, more drought-tolerant and productive. If too densely spaced, these plants crowd each other and inhibit plant development. Plant size and vigor is also important for competition with weed species. Since several of the native grasses spread by rhizomes, the stand density will improve until limited by the shade and competition of the taller plants. When we consider these objectives, a seedling density of 10 to 20 cool-season grass plants per square foot is an acceptable target.

The tall, warm-season grasses are larger and more robust (after the establishment period) than are most cool-season grasses. They also have deeper root systems. The preceding information about cool-season grasses applies even more to the warm-season grasses. Mature stands of the tall, warm-season grasses can develop full stand density with only one seedling per square foot (10 per m^2). In the Northeast, frost heaving is a problem in stands of these grasses on soils that is less than well drained. Therefore, the time to declare success is not at the end of the first growing season, but at the beginning or middle of the second growing season (June – July of the second year). Frost heaving does not appear to be a problem in stands of warm-season grasses that are older than 1.5 years. **If a warm-season grass stand has a strong plant per square foot in June of the second year, the stand will be successful in most cases.**

The relative germination and seedling vigor of different species within the warm-season grass group is worth mentioning here. Coastal panicgrass and switchgrass have the strongest vigor, and little bluestem and indiangrass have the least vigor. When evaluating mixed plantings, it is common to see few indiangrass seedlings. These tend to become visible in the second or third years and usually remain as a minor part of the stand.

Plantings for erosion control and wildlife cover often have forbs or herbaceous legumes included in the seed mix. These seed must be planted in much smaller numbers than the grasses. The reason for this is that the grasses are providing the bulk of the onsite benefits for erosion control and cover.

Stand maintenance

While native plantings may be considered permanent, periodic management is required. Management actions vary with soil, climate, plant species, and other factors. Management needs to occur before stand vigor declines dramatically, or competitive invasive species overrun a planting. A program of systematic monitoring of stand vigor is recommended to guide management decisions.

Management tools for fully established stands are the same as those for establishing stands, but vary in their application. Timing of mowing, herbicide applications, grazing, or burning can vary depending on objectives.

Management treatments on either planted or naturally occurring native sites may be undertaken for a variety of reasons. Chief among these is the removal of accumulated plant litter (which can impede light penetration) and the control of unwanted species. Experience indicates that a 2- to 3-inch (6–8 cm) layer of plant litter can reduce seed culm and total culm densities. These features are indicators of stand vigor. Exposing growth points to sunlight and recycling nutrients tied up in old plant growth with a controlled burn generally stimulates vigorous new growth.

Properly timed management, especially a properly timed burn, can effectively control invading woody plants. In a warm-season grass planting, a burn in the spring of the second or third year after establishment when the grasses have 1 to 3 inches of new growth is strongly recommended as an initial management treatment. Fire management also serves to reduce the risk of large and potentially damaging wildfires by removing accumulations of old growth.

Planned, well-controlled fire is a useful and inexpensive management technique in warm-season grass stands. Unplanned, uncontrolled fire is obviously dangerous and becomes more likely as the number of public users increases. Few people have experience with tall-grass fires that involve stands with several years of fuel built up. Therefore, the hazard should be managed with planned, cool-season grass firebreaks, and the relative need for these firebreaks should be considered in the site management process. Distances considered safe for fire may be shorter than those for the associated smoke. Smoke damage to property or smoke inhalation by humans or livestock could be a costly situation. This is especially true if one is considering a roadside site. Traffic management, posting, and permits need to be addressed before burning.

Timing, weather, moisture conditions, and firing techniques are important factors influencing the effectiveness of a managed burn. Burning research in Kansas and other States has found that forbs react differently to fire than do the grasses, and may be injured by spring burns. If the burn is intended to control shrubs and saplings, timing is critically important. Research at several universities has shown that the most effective time to injure woody plants with fire is just as they reach full leaf. At that stage, they have expended large energy reserves to create new growth and have not yet been able to replenish their carbohydrate stores through photosynthesis and respiration. Warm-season grasses will most likely have achieved more than 1 to 3 inches of new growth—the stage at which fire is most beneficial to them—when woody plants reach full leaf. However, warm-season grass vigor is not seriously affected as long as burns are not done every year.

Careful management is required if controlled burns are being used to maintain a savanna-type plant community where trees are interspersed throughout the grassland. Before undertaking any burn, consult with experts and develop a burn plan.

Mowing and grazing can provide many benefits similar to burning. Mowing should be delayed until after most ground nesting birds have completed incubation and left their nest sites. Cut as low as possible with a mower conditioner or a flail-type mower. Remove as much of the old plant litter as possible to stimulate new growth. Experience suggests that mowing does not provide a long-lasting treatment effect if the lower litter layer is not removed. If mowing or haying do not provide sufficient impact on old plant litter, scarification of the soil surface with heavy harrows or similar equipment may enhance the treatment effect. Soil surface disturbance can invite invasion by unwanted plants however.

Grazing is also a management option. In wildlife priority areas, grazing must be well-regulated, infrequent, and intended to provide maximum benefits to the grass stand. Grazing should be designed to maximize stand vigor, with agricultural benefits secondary. Extensive reclamation areas or areas of existing pasture revegetated with native plant material can be maintained in a productive state and provide nutritious long-lived forage under a managed grazing system. Local pasture experts should be consulted to set up a system that is appropriate for your soil and climate zone.

Herbicide treatments of established grass stands are often carried out as spot applications. Confining treatment to relatively small sections of the grassland can avoid unintended damage to desired species or limit it.

Combinations of treatment methods may be necessary to effectively manage grasslands associated with wetlands.

Literature cited

Breitbach, D., and L. Pollard. 1987. Establishment of warm-season grasses. USDA, Soil Conserv. Serv., Minnesota Tech. Note, Agronomy No. 3, 8 pp.

Dickerson, J., B. Wark, D. Burgdorf, A. Bush, C. Miller, R. Maher, and W. Poole. 1997. Vegetating with native grasses in Northeastern North America. USDA, Nat. Resourc. Conserv. Serv. and Ducks Unlimited Canada, 62 pp.

Duebbert, H.F., E.T. Jacobson, K.F. Higgins, and E.B. Podoll. 1981. Establishment of seeded grasslands for wildlife habitat in the prairie pothole region. USDI, Fish and Wildlf. Serv., Spec. Sci. Rep., Wildlife No. 234. 21 pp.

Masters, R.A., S.J. Nissen, R.E. Gaussoin, D.D. Beran, and R.N. Stougaard. 1996. Imidazolinone herbicides improve restoration of Great Plains grasslands. Weed Technology 10:392–403.

Missouri Conservation Commission. 1980. Native grasses for wildlife. Dep. Conserv., 4 pp.

Mlot, C. 1990. Restoring the prairie. Bio Science 40:11, pp. 804–809.

Ries, R.E., R.S. White, and R.J. Lorenz. 1987. Establishment of range plants in the Northern Great Plains. *In* J.E. Mitchell, ed., Impacts of the Conservation Reserve Program in the Great Plains. USDA For. Serv. Tech. Rep. GTR RM–148, pp. 29–34

Schramm, P. 1978. The dos and don'ts of prairie restoration. Proc., Fifth Midwest Prairie Conf., Iowa State Univ., pp. 139–144.

Sharp, W.C. Undated. Use and management of tall growing warm-season grasses in the northeast. USDA, Soil Conserv. Serv. (now NRCS), Chester, PA.

United States Department of Agriculture, Natural Resources Conservation Service. 1997. Conservation plant sheets for the Northeast United States. Plant Materials Program, East Region, No. 1–95.

Wark, D.B., W.R. Poole, R.G. Arnott, L.R. Moats, and L. Wetter. 1995. Revegetating with native grasses. Ducks Unlimited Canada, 133 pp.

Whalley, R.D.B., C.M. McKell, and L.R. Green. 1966. Seedling vigor and the non-photosynthetic stages of seedling growth in grasses. Crop Sci. 6:147–150.

III.J.1 Noxious, invasive, and alien plant species: a challenge in wetland restoration and enhancement

(Norman C. Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001)

Purpose

The purpose of this paper is to provide information on noxious, invasive, alien and other problem plant species that threaten the success of wetland restoration and enhancement projects. This paper defines the different categories of problem species, identifies the threats to success caused by these species, recommends methods of avoidance through planning and monitoring, and lists numerous species that negatively impact the function and value of wetland restoration and enhancement projects.

Contents

Noxious, invasive, other problem plant species, and the threat to wetland restoration and enhancement—Noxious and invasive plant species threaten the success of many wetland restoration and enhancement activities. When these species become established on a developing site, they can out-compete and displace native species, reduce wildlife habitat potential, alter natural ecosystem processes, and limit overall biodiversity. Although no site is immune from the chance dispersal of problem species, some sites are more predisposed than others to an invasion because of their position on the landscape, proximity to existing propagule sources, poor structural design of the project, or poor establishment of targeted vegetation. It is important to consider a site's risk to invasion in the planning process and to perform regular followup monitoring to identify chance introductions. With early detection, a problem species is easier to contain or eradicate than when it is fully established. For once it is established, it may be difficult or impossible to control or eliminate, and often attempts to do so adversely impact remnant native vegetation.

Problem plant species definitions

Weed—The USDA Animal and Plant Health Inspection Service (APHIS) defines a weed as any plant that poses a major threat to agriculture and/or natural ecosystems within the United States. Although this is more specific than a typical dictionary definition (i.e., a plant growing in an undesired location), it more accurately portrays the economic and ecological impact weeds can cause on landscapes and on the natural ecosystems being recreated. In restoration and enhancement work, species other than those listed on the APHIS Noxious Species List or State lists can (and do) limit success and must not be excluded from consideration.

Alien species—A species introduced and occurring in locations beyond its known historical range. This included introductions from other continents, bioregions, and also those not native to the local geographic region. Executive Order (E.O.), Invasive Species, February 3, 1999, more narrowly defines an alien species and ties the definition to an occurrence outside a native vegetation. That definition is "alien species means, with respect to a particular ecosystem, any species, including its seeds, eggs, spores, or other biological material capable of propagating that species, that is not native to that ecosystem." Synonyms for alien species include exotic, non-native, nonindigenous, and introduced species. Of the thousands of plants that have been introduced to the United States intentionally for cultivation or by accident, about 4,000 of these alien plant species now occur outside of cultivation. Only about 400 of these are considered problematic with respect to adverse effects on agricultural or native biota. For example, saltmarsh cordgrass (*Spartina alterniflora*) is native to eastern North American estuaries. It has been introduced to western North American shoreline habitats and, there it is considered an alien species. In these western habitats, saltmarsh cordgrass adversely impacts native habitats and displaces native plant species.

Native species—As defined in E.O., Invasive Species, February 3, 1999, a "native species means, with respect to a particular ecosystem, a species that, other than as a result of an introduction, historically occurred or currently occurs in that ecosystem." Accordingly, a species cannot be considered native to a geographic region or habitat merely because it occurs natively somewhere within the continental United States. For example, pond pine (*Pinus serotina*) occurs natively in wetlands in the Southeast and Mid-Atlantic States from Alabama to New Jersey. Although habitats may be similar in the Lower Mississippi River Alluvial Valley (LMRAV), pond pine cannot be considered native to this geographical region because its distribution does not extend natively into the LMRAV. Another example, coastal doghobble (*Leucothoe axillaris*) occurs in Coastal Plain wetlands throughout the Southeast. It would not be considered native to montane wetlands of the same Southeastern states since these habitats are different from those on the Coastal Plain.

Invasive species—A species that demonstrates rapid growth and spread, invades habitats, and displaces other species. Species that are prolific seed producers, have high seed germination rates, easily propagated asexually by root or stem fragments, and/or rapidly mature predispose a plant to being an invasive. For example, the hybrid cattail (*Typha* x *glauca*), a cross between native cattails, is extremely aggressive and out-competes its parents and other native species when established. Alien species that are predisposed to invasiveness have the added advantage of being relatively free from predators (herbivores, parasites, and disease) and can, therefore, expend more energy for growth and reproduction. For example, Nepal microstegium (*Microstegium vimineum*) introduced from Asia displaces native vegetation on flood plains and other moist environments creating a monoculture in the herbaceous layer. *Microstegium* now occurs in 21 States and Puerto Rico, ranging from Texas to Florida in the South and north into New York and Illinois.

Noxious species—Any living stage (including but not limited to seeds and reproductive parts) of any parasitic or other plant of a kind, or subdivision of a kind, which is of foreign origin, is new to or not widely prevalent in the United States, and can directly or indirectly injure crops, other useful plants, livestock, or poultry or other interests of agriculture, including irrigation, or navigation or the fish and wildlife resources of the United States or the public health (Federal Noxious Weed Act). For example, purple loosestrife (*Lythrum salicaria*) was introduced into the Northeast in the early 1800s and now occurs in wetlands in 41 of the 48 conterminous United States. It aggressively develops dense monocultural stands, degrades native vegetation, reduces overall vegetative biodiversity, and directly impacts wildlife by the loss of habitat and food.

Noxious, invasive, and alien species effect on wetland restoration and enhancement success

Noxious, invasive, and alien species have been referred to as a form of biological pollution because they can upset the balance among native species within natural and agricultural ecosystems (USDA, APHIS). It must be reiterated that alien does not equal bad; not all alien species are invasive or become noxious. In fact, many non-native species peacefully coexist with native vegetative ecosystems and provide habitat support for native wildlife and the human population. The bulk of our agricultural food crops are of alien origin. However, there are numerous examples of aliens that paint a different picture and pose significant threats to the natural and agricultural landscapes and their biota. Although each alien, invasive, and noxious species acts differently in the environment, and in its interaction with other species, several patterns of disruptive behavior occur. Some of the more common ways these species affect the native biota follow.

Replacement of native vegetation systems—Alien species commonly have few natural predators, parasites, or diseases and can simply out-compete native species, such as leafy spurge (*Euphorbia esula*).

Reduction of biodiversity—One invasive species can rapidly out-compete numerous native species and dominate an ecosystem with its accentuated reproductive potential. This creates a less diverse flora. The Nature Conservancy estimates that alien, invasive species are implicated in 42 percent of the species listed as endangered or threatened under the Endangered Species Act.

Reduction of wildlife habitat and food—Native plants (providing food and habitat) and native animals have co-evolved over long periods. Replacement of native vegetative systems to ones dominated by aliens generally alters these co-evolved relationships. For example, reed canarygrass (*Phalaris arundinacea*) establishes itself in dense monocultural stands in restored and enhanced wetlands throughout the Northeast, Great Plains, and Pacific Northwest regions and offers little wildlife food value in the seed. An example of how altered vegetation can affect habitat structure can be demonstrated with purple loosestrife (*Lythrum salicaria*). It replaces annual emergent vegetation with its perennial subwoody stems in wetlands throughout much of the United States and Canada. In contrast to these examples, some aliens are planted to provide specific benefits for specific species. In such cases the net benefits provided to the targeted species overrides the cost to the native vegetation and other wildlife species. For example, Japanese millet (*Echinochloa crus-galli* var. *frumentacea*), an alien annual introduced from Asia, is commonly planted in waterfowl management areas for its prolific seed production and forage value.

Change ecosystem processes—Native ecosystems have developed under particular abiotic factors and ecosystem processes (e.g., rainfall patterns, fire regimes, rates of nutrient cycling) and have adapted to them. The presence of some alien species alter these processes, which alters the ecosystem to where it can no longer support the native vegetative or faunal community. Flack and Benton (1998) list several important examples. Paper bark tree (*Melaleuca quinquenervia*) is invading herbaceous marshes in southern Florida and converts these habitats to woody dominated swamp forests. Another example is tamarisk (*Tamarix gallica*). It has been introduced into the South and Southwest and alters the natural hydrologic cycle by transpiring greater quantities of water than native vegetation in the same habitat, thus reducing water tables and some surface water habitats. Tamarisk also concentrates salt in its leaves, and the decomposing leaf litter raises the site salinity.

Hybridization—Hybridization between species and populations affects native floras in many ways. Our activities often bring two similar species together and they hybridize. When this occurs the resulting offspring may not be as "fit" to survive as either parent (e.g., oak hybrids). On the contrary, there are incidences where the opposite occurs when two species are brought together, hybridize, and the hybrid is more competitive than either parent is. For example, the hybrid cattail (*Typha* x *glauca*) is a cross between the broadleaf (*T. latifolia*) and narrowleaf (*T. angustifolia*) cattails and between broadleaf and southern (*T. domingensis*) cattails. Barkely (1986) cites the hybrid as "developing extensive pure stands by rhizomatous growth" where it occurs in native prairie marshes with greatly varying water levels and in disturbed habitats. In these habitats the hybrid can out-compete its parents as well as other native species. For example, Susan Galatowitsch (1994) reports that the hybrid cattail has replaced native white top (*Scolochloa festucacea*) and wild rice (*Ziziania aquatica*) in prairie potholes in northern Iowa.

Another example of hybridization affecting native species and populations can be drawn from attempts to restore natural habitats using native seed and plant materials, but the originating source (genetic stock) is from a nonlocal source. It is well known by horticulturists and plant materials specialists that populations of plants adapt to their local environment over many generations. When these locally adapted populations are grown outside of those conditions, they are often less well adapted. When these introduced natives hybridize with the local species, the genetic makeup of the locals become altered and, possibly, less well adapted to their own local environment. This type of hybridization, which can result in lowered fitness of local populations, is of critical concern when rare, endangered, and threatened species are involved.

Mitigation of the effects

Restoration and enhancement activities can significantly contribute to the spread of noxious, invasive, and alien species and the detrimental effects caused by them. The colonization and establishment of these problem species on a site can affect the overall success of a project and can inadvertently create additional opportunities for the establishment and spread of these species throughout the watershed and onto other landscapes and land ownerships. Although it may be impossible to eliminate all possibilities of invasion, their presence, impact, and spread can be limited through proper planning and monitoring.

Know the species that can cause problems in the area. State heritage programs, the Nature Conservancy, Extension Service, APHIS, and other state and private programs maintain lists of problem species. Request and maintain these lists. Be familiar with the species' identification, method of dispersal, and time of year it occurs and reproduces. Check the NRCS Plant Data Center's PLANT Web site for the weed module.

Be sensitive to the threat these species pose. Often specific alien and rapidly proliferating plant species are used to enhance particular wetland functions. Understand the potential of these species for colonizing surrounding landscapes and the possible effects they may produce outside the project site. The targeted vegetation may need to be altered if the threat to surrounding landscapes and properties is too great.

Plan accordingly. In the planning process, identify potential sources of accidental introduction onto a project site. Are particular problem species on adjacent properties, upstream, or upwind? Are they capable of migrating to the project site?

Detect the presence of propagules in seed banks, mulch, equipment, or weed seed contained in planted seed (e.g., Canada Thistle as a contaminant in native warm-season grass seed). If seed or other propagules of problem species are known (or suspected) to be in the seed bank in high concentrations, it may be advisable to delay the installation of the restoration for one growing season and concentrate on pest control.

Develop monitoring protocols. Monitoring protocols should stress identification, early detection, and control/eradication. Knowing potential problem species in the area and their presence in the local landscape can be used to determine the rigor of monitoring necessary.

Be knowledgeable in the approved methods of control (cultural, mechanical, chemical, biological) for the species. Often invading species can be controlled mechanically by hand removal if detected early and before they have a chance to reproduce. The chemical pesticides and biological agents available for use on invasive and noxious species varies by state. Contact County Extension, State agriculture departments or State pest management departments for approved materials and techniques. For chemical control, identify the specific herbicides recommended, rates of application, mixing instructions, special application techniques, and the timing of application for most effective control. When using chemical methods, be aware of the impact the herbicides have on existing native species. If recommending approved biological controls, be aware of similar types of information including kind of biological agent to be used, timing of release, duration of impact, intensity of activity of the biological agent, and any special precautions or requirements.

Avoid establishing non-native and tame species where possible. For example, fescues, bermudagrasses, bahiagrass, and brome grasses are commonly planted because they are available, inexpensive, and easily established. These species do provide quick cover, but their benefits are short-term, have limited wildlife benefit, and will restrict overall site biodiversity by developing dense, persistent monocultural stands. Native species may not give the visual appearance of quick success; however, for the long-term they provide greater species diversity and wildlife habitat.

Purchase and plant native species that were derived from local collections. When active regeneration (i.e., planting) is used as a technique in vegetative regeneration, specify that the plant materials are to be derived from local locations. Just because the vendor is local, does not mean that the material originated locally. Native, locally adapted vegetation generally outperforms nonadapted strains of the same species. The result will be a quicker establishment of cover on the restoration site.

Reestablish vegetation on a disturbed site quickly. Many invasive and noxious species are rapid colonizers of bare soil. Restoration and enhancement sites that are allowed to revegetate naturally, but do not have realistic sources of propagules in the soil and/or adjacent landscapes are exceptionally vulnerable to infestation of problem species. To repeat an old agricultural cliché, "For land's sake, keep it covered."

Most common invasive and noxious plant species colonizing wetland restoration and enhancement sites

The following list of species is known to invade and colonize restored and enhanced wetland sites. This list is by no means exhaustive, and one should be familiar with other species in their area. Methods of identification and control of these and other species can be obtained from County Extension, State, private organizations, and on Internet Web sites. The Wetland Science Institute will be issuing individual treatments on many of these species in future issues of Wetland Restoration and Enhancement Technical Series. These treatments of individual noxious and invasive species will include information on field identification, distribution within North America, detrimental effects caused by the species, reproductive and dispersal strategies, and other aspects of the species life history that may help in control. These technical notes will have general information on established methods of control (cultural, mechanical, chemical, biological) that can be locally refined.

- Canada thistle (*Cirsium arvense*)
- Chinese tallow (*Sapium sebiferum*)
- Giant reed (*Arundo donax*)
- Hybrid cattail (*Typha* x *glauca*)
- Leafy spurge (*Euphorbia esula*)
- Paper bark tree (*Melaleuca quinquenervia*)
- Phragmites (*Phragmites australis*)
- Purple loosestrife (*Lythrum salicaria*)
- Reed canarygrass (*Phalaris arundinacea*)
- Tamarisk (*Tamarix gallica*)

Useful Internet Web sites for additional information on invasive and noxious species

NRCS Plant Data Center's weed module; Federal Noxious Weed List; and State noxious weed lists are on the USDA-NRCS-PLANTS Data Center's Web site :
plants.usda.gov

Alien Plant Working Group:
www.nps.gov/plants/alien

The Nature Conservancy Wildland Weeds Management and Research Program:
tncweeds.ucdavis.edu/esadocs.html

Vegetation Management Guidelines:
www.inhs.uiuc.edu/edu/VMG/VMG.html

Southeast Exotic Pest Plant Council:
www.se-eppc.org

Extoxnet information on herbicides:
ace.orst.edu/info/extoxnet

References

Barkley, Ted (ed.). 1986. Flora of the Great Plains.

Clinton, William J. 1999. Invasive species. Executive Order, Feb. 3.

Federal Noxious Weed Act of 1974. 7 U.S.C. 2809.

Flack, Stephanie R., and Nancy B. Benton. 1998. National Wetlands Newsletter (May-June) 20–3, p. 7–11.

Galatowitsch, Susan, and Arnold van der Valk. 1994. Restoring prairie wetlands, an ecological approach. Iowa State Univ. Press, Ames, Iowa, 246 pp.

Federal Noxious Weed List

(June 7, 1999)

Aquatic/Wetland

Azolla pinnata (Azollaceae) (mosquito fern, water velvet)
Caulerpa taxifolia (Caulerpaceae)(Mediterranean clone of caulerpa)
Eichhornia azurea (Ponterderiaceae) (anchored waterhyacinth)
Hydrilla verticillata (Hydrocharitaceae) (hydrilla)
Hygrophila polysperma (Acanthaceae) (Miramar weed)
Ipomoea aquatica (Convolvulaceae) (Chinese waterspinach)
Lagarosiphon major (Hydrocharitaceae) (Oxygen weed)
Limnophila sessiliflora (Scrophulariaceae) (ambulia)
Melaleuca quinquenervia (Myrtaceae) (melaleuca)
Monochoria hastata (Pontederiaceae) (monochoria)
Monochoria vaginalis (Pontederiaceae) (pickerel weed)
Ottelia alismoides (Hydrocharitaceae) (duck-lettuce)
Sagittaria sagittifolia (Alismataceae) (arrowhead)
Salvinia auriculata (Salviniaceae) (giant salvinia)
Salvinia biloba (Salviniaceae) (giant salvinia)
Salvinia herzogii (Salviniaceae) (giant salvinia)
Salvinia molesta (Salviniaceae) (giant salvinia)
Solanum tampicense (Solanaceae)(wetland nightshade)
Sparganium erectum (Sparganiaceae) (exotic bur-reed)

Parasitic

Aeginetia spp. (Orobanchaceae)
Alectra spp. (Scrophulariaceae)
Cuscuta spp. other than native or widely distributed species (Cuscutaceae)(dodders)
Orobanche spp. other than native or widely distributed species (Orobanchaceae) (broomrapes)
Striga spp. (Scrophulariaceae) (witchweeds)

Terrestrial

Ageratina adenophora (Asteraceae) (crofton weed)
Alternanthera sessilis (Amaranthaceae) (sessile joyweed)
Asphodelus fistulosus (Liliaceae) (onionweed)
Avena sterilis L. (Poaceae) (animated or wild oat)
Spermacoce alata (Rubiaceae) (borreria)
Carthamus oxyacanthus (Asteraceae) (wild safflower)
Chrysopogon aciculatus (Poaceae) (pilipiliula)
Commelina benghalensis (Commelinaceae) (Benghal dayflower)
Crupina vulgaris (Asteraceae) (common crupina)
Digitaria abyssinica (=D. scalarum) (Poaceae) (African couch grass)
Digitaria velutina (Poaceae) (velvet fingergrass)
Drymaria arenarioides (Caryophyllaceae) (lightening weed, alfombrilla)

Federal Noxious Weed List—Continued

Emex australis (Polygonaceae) (three-cornered jack)
Emex spinosa (Polygonaceae) (devil's thorn)
Galega officinalis (Fabaceae) (goatsrue)
Heracleum mantegazzianum (Apiaceae) (giant hogweed)
Imperata brasiliensis (Poaceae) (Brazilian satintail)
Imperata cylindrica (Poaceae) (cogongrass)
Ischaemum rugosum (Poaceae) (murain-grass)
Leptochloa chinensis (Poaceae) (Asian sprangletop)
Lycium ferocissimum (Solanaceae) (African boxthorn)
Melastoma malabathricum (Melastomataceae) (no common name)
Mikania cordata (Asteraceae) (mile-a-minute)
Mikania micrantha (Asteraceae) (mile-a-minute)
Mimosa invisa (Fabaceae) (giant sensitive plant)
Mimosa pigra (Fabaceae) (catclaw mimosa)
Nassella trichotoma (Poaceae) (serrated tussock)
Opuntia aurantiaca (Cactaceae) (jointed prickly pear)
Oryza longistaminata (Poaceae) (red rice)
Oryza punctata (Poaceae) (red rice)
Oryza rufipogon (Poaceae) (red rice)
Paspalum scrobiculatum (Poaceae) (Kodo-millet)
Pennisetum clandestinum (Poaceae) (kikuyugrass)
Pennisetum macrourum (Poaceae) (African feathergrass)
Pennisetum pedicellatum (Poaceae) (kyasuma-grass)
Pennisetum polystachion (Poaceae) (missiongrass)
Prosopis alapataco (Fabaceae) (*Prosopis* spp. are mesquites)
Prosopis argentina
Prosopis articulata
Prosopis burkartii
Prosopis caldenia
Prosopis calingastana
Prosopis campestris
Prosopis castellanosii
Prosopis denudans
Prosopis elata
Prosopis farcta
Prosopis ferox
Prosopis fiebrigii
Prosopis hassleri
Prosopis humilis
Prosopis kuntzei
Prosopis pallida
Prosopis palmeri
Prosopis reptans
Prosopis rojasiana
Prosopis ruizlealii
Prosopis ruscifolia
Prosopis sericantha
Prosopis strombulifera
Prosopis torquata

Federal Noxious Weed List—Continued

Rottboellia cochinchinensis (Poaceae) (itchgrass)
Rubus fruticosus (Rosaceae) (wild blackberry complex)
Rubus moluccanus (Rosaceae) (wild blackberry)
Saccharum spontaneum (Poaceae) (wild sugarcane)
Salsola vermiculata (Chenopodiaceae) (wormleaf salsola)
Setaria pallide-fusca (Poaceae) (cattail grass)
Solanum torvum (Solanaceae) (turkeyberry)
Solanum viarum (Solanaceae) (tropical soda apple)
Tridax procumbens (Asteraceae) (coat buttons)
Urochloa panicoides (Poaceae) (liverseed grass)

Added by act of Congress (H.R.2160.30, Sec 728)

Pueraria lobata (Fabaceae)(kudzu)

III.J.2.a Management and control of Canada thistle (*Cirsium arvense* (L.) Scop.) (CIAR4)

(Victoria Nuzzo, Native Landscapes, Rockford, Illinois, original abstract 1997; John M. Randall, The Nature Conservancy, Arlington, Virginia, revised 1998; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; State Distribution Maps and Photographs, NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

The purpose of this paper is to provide information on the biology and control of invasive plant species in wetlands.

To the user

Element Stewardship Abstracts (ESAs) are prepared to provide The Nature Conservancy's Stewardship staff and other land managers with management-related information on those species and communities that are most important to protect or most important to control. The abstracts organize and summarize data from numerous sources including literature and researchers and managers actively working with the species or community.

We hope, by providing this abstract free of charge, to encourage users to contribute their information to the abstract. This sharing of information will benefit all land managers by ensuring the availability of an abstract that contains up-to-date information on management techniques and knowledgeable contacts. Contributors of information will be acknowledged within the abstract and receive updated editions.

For ease of update and retrievability, the abstracts are stored on computer at the national office of The Nature Conservancy. Each abstract has a Nature Conservancy office or program responsible for its updating and maintenance. The address and telephone number of the office is recorded on the first page of the abstract. Anyone with comments, questions, or information on current or past monitoring, research, or management programs for the species or community described in an abstract should contact the Land Steward in the office responsible for that abstract.

This abstract is a compilation of available information and is not an endorsement of particular practices or products.

Please do not remove this cover statement from the attached abstract.

1815 North Lynn Street, Arlington, Virginia 22209 (703) 841 5300

Content

The Nature Conservancy
Element Stewardship Abstract

Canada Thistle
Creeping Thistle
Californian Thistle

Scientific name

Cirsium arvense (L.) Scop. Early nomenclature is recorded in Detmers (1927).

Common name

Canada thistle is the common name used in the United States and Canada. Outside North America the plant is also referred to as creeping thistle and Californian thistle (Jessep 1989).

Diagnostic characters

Cirsium arvense is an erect perennial rhizomatous thistle, usually 1.5 to 3.0 feet tall, distinguished from all other thistles by creeping horizontal lateral roots, dense clonal growth, and small dioecious (male and female flowers on separate plants) flowerheads. Four varieties are recognized: var. *vestitum* Wimm. and Grab. (leaves gray-tomentose below), var. *integrifolium* Wimm. and Grab. (leaves glabrous below, thin, flat, and entire or shallowly pinnatifid), var. *arvense* (leaves glabrous below, thin, flat, and shallowly to deeply pinnatifid), var. *horridum* Wimm. and Grab. (leaves glabrous below, thick and wavy, with many marginal spines) (Moore 1975). The most common variety of the species in North America is *horridum*. All varieties are interfertile, and one plant of var. *integrifolium* produced seedlings of all four varieties (Detmers 1927). Within each variety, numerous genotypes vary in appearance and in response to management activities. Additionally, *Cirsium arvense* changes morphology in response to environmental conditions (Nadeau and Vanden Born 1989).

Chromosome number for all *Cirsium arvense* varieties is $2n = 34$ (Moore and Frankton 1974). There are approximately 350 species worldwide in the genus *Cirsium* (Moore and Frankton 1974).

Cirsium arvense can be confused with other thistles, especially bull thistle (*Cirsium vulgare*), and the closely related musk thistles (*Carduus* sp). Distinguishing characteristics of *Cirsium arvense* are flowerheads small (<1 inch high) and dioecious and stems not conspicuously spiny-winged (Moore and Frankton 1974). All species of *Cirsium* (plumed thistle) have a pappus with branched hairs, in contrast to the unbranched pappus hairs on *Carduus* (plumeless thistle) (Moore 1975).

Phenology of *Cirsium arvense* varies with ecotype, but follows a general pattern. In Washington State, overwintering Canada thistle roots develop new underground roots and shoots in January and begin to elongate in February (Rogers 1928). Shoots emerge March to May when mean weekly temperatures reach 5 degrees Celsius. Rosette formation follows, with a period of active vertical growth (about 1.25 in/d) in mid-to-late June. Flowering is from June to August in the U.S., and June to September in Canada, when days are 14 to 18 hours long (Hodgson 1968, Van Bruggan 1976, Moore 1975). *Cirsium arvense* is a long-day plant (Linck and Kommendahl 1958, Hunter and Smith 1972).

Cirsium arvense illustrations are in Detmers (1927), Rogers (1928), and Haderlie et al. (1987).

Stewardship summary

Despite its common name, Canada thistle is native to Europe and was apparently introduced to North America in the early 17th century (Hansen 1918). *Cirsium arvense* was declared a noxious weed by the State of Vermont in 1795 (Hansen 1918). By 1918, it was on the noxious weed lists of 25 Northern States, and by 1991, it had been declared noxious by at least 35 States and 6 Canadian Provinces (Moore 1975). It is now widespread in all U.S. States and Canadian Provinces between 37 and 58 to 59 degrees N (Moore 1975).

Cirsium arvense is invasive in prairies and other grasslands in the Midwest and Great Plains and in riparian areas in the Intermountain West. It is particularly troublesome in the Northwest and Northcentral States, and in Southern Canada (Moore 1975). Canada thistle spreads primarily by vegetative means and secondarily by seed.

Cirsium arvense has numerous ecotypes that respond differently to management activities. Some infestations may be completely controlled by one technique, while others are only partly controlled because two or more ecotypes are present within the population. Additionally, *Cirsium arvense* responds differently to management under different weather conditions. Therefore, it is often necessary to implement several control techniques and to continuously monitor their impacts.

Where possible, all *Cirsium arvense* plants within a site should be killed. Where resources are limited, two strategies are recommended:

- Target *Cirsium arvense* clones based on location, controlling plants in high quality areas first, then in low quality areas. Treat entire clones to prevent resprouting from undamaged roots.
- Target female clones to reduce seed production and additional spread of *Cirsium arvense*. However, some apparently "male" clones are self-fertile.

Control techniques for natural areas are constrained by the need to minimize damage to native species. The best option in prairies and other grasslands is to first enhance growth of native herbaceous species by spring burning, and then cut or spot treat Canada thistle with glyphosate when it is in late bud or early bloom (usually June). It is necessary to prevent shoot growth for at least 2 years to deplete roots and kill Canada thistle. Several biological control agents have been released against Canada thistle, but overall they provide little or no control at the population level although they may weaken and kill individual plants.

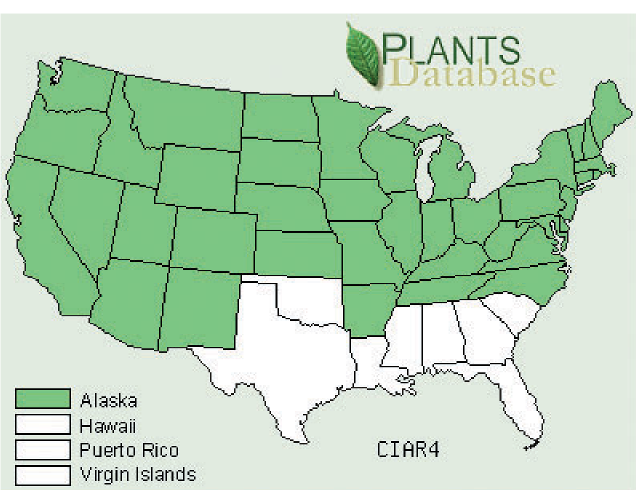
Threats posed by this species

Natural areas invaded by Cirsium arvense include prairies and other grasslands in the Midwest and Great Plains and riparian areas in the Intermountain West. *Cirsium arvense* threatens natural communities by directly competing with and displacing native vegetation, decreasing species diversity, and changing the structure and composition of some habitats. Species diversity in an undisturbed Colorado grassland was inversely proportional to the relative frequency of Canada thistle (Stachion and Zimdahl 1980). Canada thistle invades natural communities primarily through vegetative expansion and secondarily through seedling establishment.

Cirsium arvense presents an economic threat to farmers and ranchers. Infestations reduce crop yield through competition for water, nutrients, and minerals (Malicki and Berbeciowa 1986) and interfere with harvest (Boldt 1981). In Canada, the major impact of *Cirsium arvense* is in agricultural land and in natural areas that have been disturbed or are undergoing restoration (White, et al. 1993). In the U.S., it is a host for bean aphid and stalk borer, insects that affect corn and tomatoes (Moore 1975), and for sod-web worm (Crampus sp.), which damages corn (Detmers 1927). In Bulgaria, *Cirsium arvense* is a host for the cucumber mosaic virus (Dikova 1989). In addition to reducing forage and pasture production, Canada thistle may scratch grazing animals, resulting in small infections (Moore 1975).

Range

Cirsium arvense is native to Southeastern Europe and the Eastern Mediterranean (Moore 1975) and possibly to Northern Europe, Western Asia, and Northern Africa (Detmers 1927, Amor and Harris 1974). It now has a near global distribution between 37 and 58 to 59 degrees N in the Northern Hemisphere (Moore 1975), and at latitudes greater than 37 degrees S in the Southern Hemisphere exclusive of Antarctica (Amor and Harris 1974). *Cirsium arvense* occurs throughout Europe, Northern Africa, Western and Central Asia, Northern India, Japan, China, and Northern North America, South Africa, New Zealand, Tasmania, and Southeastern Australia (Dewey 1991, Rogers 1928, Hayden 1934, Amor and Harris 1974).



In 1975, Canada thistle's range was an estimated 3.8 million square miles in North America, extending over an area 1.300 miles north to south, and 3,000 miles east to west (Moore 1975). *Cirsium arvense* infestations here are particularly troublesome in the Northwest and Northcentral States, especially north of the 35th parallel (Dewey 1991), and in the Southern Canada (Moore 1975). The species range is determined by rainfall, temperature, and day length. The northern limit of the zone of highest density in Canada corresponds with the –18 degrees Celsius (0 °F) mean January isotherm; whereas, the southern limit of the species is probably controlled by high summer temperatures and short-day length (Moore 1975).

Optimal growth occurs at 77 degrees Fahrenheit day and 59 degrees Fahrenheit night, in mesic soil with high nitrogen (15–30 ppm) (Haderlie, et al. 1987). In Montana, the plant grows best where rainfall averages 50 to 75 centimeters per year (Hodgson 1968), while in Australia, the heaviest infestation occurs where annual rainfall averages 70 to 100 centimeters (Amor and Harris 1974).

Habitat

Cirsium arvense occurs in nearly every upland herbaceous community within its range and is a particular threat in prairie communities and riparian habitats. In the Great Plains, Canada thistle invades wet and wet-mesic grasslands as well as prairie potholes in the Dakotas. It also invades riparian areas and along irrigation ditches from the Western Plains across the northern half of the Intermountain West to the Sierra Nevada and Cascade ranges. In the upper Midwest (Wisconsin and Illinois), *Cirsium arvense* is found in degraded sedge meadows, growing on tussocks elevated above the normal high water line. In Canada, *Cirsium arvense* is frequent in prairie marsh (Thompson and Shay 1989) and sedge meadow (Hogenbirk and Wein 1991). Throughout its range it is common on roadsides; in old fields, croplands, and pastures; and in deep, well-aerated, mesic soils. In Eastern North America, it occasionally occurs in relatively dry habitats, including sand dunes and sandy fields, as well as on the edge of wet habitat, including streambanks, lakeshores, cleared swamps, muskegs, and ditches (Moore 1975).

Canada thistle is shade intolerant. It grows along the edge of woods (both deciduous and coniferous), but is rarely found within forests.

Cirsium arvense grows on all but waterlogged, poorly aerated soils including clay, clay loam, silt loam, sandy loam, sandy clay, sand dunes, gravel, limestone, and chalk, but not peat (Rogers 1928, Korsmo 1930 [cited in Moore 1975], Bakker 1960, Hodgson 1968, Moore 1975). It grows best on mesic soils. In a transplant experiment, Hogenbirk and Wein (1991) determined that *Cirsium arvense* cover increased 5- to 13-fold when sods were moved from a wetland to a mesic location. *Cirsium arvense* can tolerate soils with up to 2 percent salt content.

Reproduction

Flowers—*Cirsium arvense* produces numerous small flowers clustered in heads that are typically 1 to 1.5 centimeters in diameter and 1.3 to 1.5 centimeters tall. Flower color ranges from lavender to pink or white. Flowering is triggered by long days. Ecotypes vary in their light requirements, with some ecotypes blooming during 16-hour days and others during 14-hour days; at shorter day lengths, flowering can be temperature dependent (Hunter and Smith 1972). Studies indicated Canada thistle required 14 hours of daylight per day to flower in South Dakota (Lym and Zollinger 1995), 15 hours of daylight per day in Nebraska (Hoefer 1981), and more than 15 hours of daylight per day in Idaho (Haderlie, et al. 1987).

The blooming period is longer in northern locales than in more southerly areas. In Canada, flowering begins mid-June to early July and continues into September (Moore 1975), while in Idaho and Montana, flowering begins early July and continues into August (Hodgson 1964, 1968).

Cirsium arvense is usually dioecious, with male and female flowers produced on separate plants. Female (pistillate) flowers can be readily distinguished from male (staminate) flowers by the absence of pollen (abundant in male flowers) and presence of a distinct vanilla-like fragrance (Rogers 1928), as well as by shorter corolla lobes (2.8 mm vs. 4.8 mm; Kay 1985). In seed, female flowers have a larger pappus (23 mm vs. 11 mm) and larger involucre (19 mm vs. 13 mm; Kay 1985).

Under good growing conditions, female plants produce an average of 29 flowering shoots per square meter, each with an average of 41 heads per shoot and 59 seeds per head (Bakker 1960). Total florets (individual flowers within each flowerhead) per plant varies by clone and can range from about 100 (Hayden 1934) to 430 to 1,120 (Lalonde and Roitberg 1989). Although traditionally considered dioecious, up to 26 percent of male plants are actually self-fertile hermaphrodites (male and female flowers on the same plant) capable of producing seeds. In Britain, 15 percent of clones with male flowers were actually hermaphrodites that produced 10 to 65 seeds per flowerhead, and an additional 11 percent of plants were subhermaphrodites that produced 2 to 10 seeds per flowerhead (Kay 1985). Hermaphrodites closely resemble typical male flowers (Kay 1985). Incidence of hermaphrodism varies by locality, and some areas have plants that are nearly or all truly dioecious (Lloyd and Myall 1976). Clones and individual stems can be imperfectly dioecious. Hodgson (1964) found that male and female flowers developed on separate stems grown from a single clone.

With the exception of hermaphrodites, *Cirsium arvense* flowers are obligate outcrossers. Flowers are almost exclusively insect-pollinated (Lalonde and Roitberg 1994). More insect species visit *Cirsium arvense* than other *Cirsium* or *Carduus* species due to the accessibility of its copious nectar (Ellis and Ellis-Adam 1992). Although *Cirsium arvense* may help maintain diversity of pollinating insects in this way (Ellis and Ellis-Adam 1992), it negatively impacts native plant communities and may thus have a negative impact on overall insect diversity as well.

Stigmas are receptive for at least 3 days when pollen is abundant and for more than 5 days when pollen availability is low (Lalonde and Roitberg 1994). Seed set in females is constrained by pollen availability and is highest when male and female plants are near each other, but decreases sharply when female plants are more than 50 meters from male plants (Lalonde and Roitberg 1994).

Seeds—Seeds (achenes) range from 2.5 to 3.2 millimeters long and average 1 millimeter in diameter (Rogers 1928). Ripe seeds have a tawny color. Seed weight varies by ecotype, ranging from 0.67 to 1.52 milligrams per seed (Hodgson 1964) and averaging 1.08 milligrams (Terpstra 1986). Mean seed weight is highest in seeds produced early in the summer and declines over the season (Lalonde and Roitberg 1989).

Seed set is highest when male and female plants are intermixed and decreases when female plants are more than 150 feet from male plants (Lalonde and Roitberg 1994). Seed formation has been documented when male and female plants are 150 to 300 feet apart (Bakker 1960, Hayden 1934), 150 to 300 feet apart (Lalonde and Roitberg 1994), 600 feet apart (Detmers 1927), and 1,300 feet apart (Amor and Harris 1974). Flowers must be open 8 to 10 days before seeds are mature enough to germinate (Derschied and Schultz 1960).

Females produce an average of 40 to 59 seeds per flowerhead (Hayden 1934, Bakker 1960, Kay 1985), and males average 6 plus or minus 4 seeds per head (Kay 1985). Seed production is much higher with insect pollination (40 to 85 seeds per head) than wind pollination (0.2 to 0.8 seeds per head). Seed production and viability is higher under full sun than low light (Bakker 1960).

A single plant produces an average of 1,500 seeds and up to 5,300 seeds (Moore 1975). Multiple plants produced an average of 100 to 64,300 viable seeds per square meters in Australia (Amor and Harris 1974) and up to 30,200 per square meters in Holland (Bakker 1960).

Seed dispersal—Seed dissemination occurs 2 to 3 weeks after pollination (Lalonde and Roitberg 1989). The pappus breaks off easily from the seed, often leaving seeds in the flowerhead. Most *Cirsium arvense* seeds apparently land near the parent plant; less than 10 percent of seeds found 10 meters from the parent plant still had a pappus attached (Bakker 1960). On the other hand, some long-distance dispersal occurs as evidenced by the 0.2 percent of seeds found with a pappus still attached 0.5 mile from the parent plant.

Cirsium arvense seeds spread as a contaminant in agricultural seeds (Rogers 1928) in hay and in cattle and horse droppings and on farm machinery. They may also be transported by water (Hope 1927).

Germination—Germination is affected by genotype, planting depth, substrate stratification, temperature, day length, and seed freshness. Germination and dormancy vary with ecotype, and some ecotypes have consistently low germination rates and/or long dormancy periods (Hodgson 1964).

Seeds germinate best at shallow depth (0.125 to 0.5 inch [Wilson 1979], 0.5 to 2.0 inches [Terpstra 1986]), but seed longevity increases with increased depth of planting. Seed viability appears to be a function of dormancy; once dormant, seeds remain viable until conditions change (Roberts and Chancellor 1979). Conditions change frequently for seeds planted at a shallow depth or in cultivated soil; hence, most seed in farm fields germinates within the first year and the remainder rapidly loses viability. Bakker (1960) determined that seed buried 0.33 inch deep lost all viability after 10 months, while seed buried 15 inches deep retained 35 to 39 percent viability after 30 months. Some buried seed remained viable for at least 21 years in the U.S. (Toole and Brown 1946) and 26 years in Denmark (Madsen 1962). Seed buried 40 inches deep had higher viability than seed buried 8 inches deep (Goss 1925 cited in Detmers 1927), and 40 percent of seeds remained viable after 30 months storage at 20-inch depth under water (Bakker 1960).

Germination rates are highest in loam and sand substrates, but are zero in rubble or turf (Bostock and Benton 1983). Optimum germination occurs at pH 5.8 to 7.0 (Wilson 1979). Seed viability is very low (0.5%) after passage through bovine digestive tracts (Lhotska and Holub 1989).

Seed germinates best at temperatures of 25 to 30 degrees Celsius (Bakker 1960, Amor and Harris 1974), but can germinate at lower temperatures in high light conditions. Young seeds germinate well in high light conditions, and old seeds in low light conditions (Kolk 1947 cited in Moore 1975).

In Australia, germination rates from 40 populations averaged 78 percent plus or minus 2 percent (range 52–97%; Amor and Harris 1974). Some seeds germinated the year they formed, but most germinated the following spring (Rogers 1928). In England more than 90 percent of germination occurs in April and May (Roberts and Chancellor 1979). Fresh seed may have low or high germination rates: Bakker (1960) reported 14 percent germination with fresh seed, 34 percent after 3 months, 44 percent after 6 months, and no germination thereafter when planted 1 centimeter deep in Holland. However, Hayden (1934) and Derschied and Schultz (1960) reported that fresh seed had the highest germination, up to 95 percent, 6-month-old seed had 10 to 27 percent viability, and 2-year-old seed had 15 to 71 percent viability.

The vast majority of germinating seeds develops into female plants (94–100%; Lalonde and Roitberg 1994).

Roots—*Cirsium arvense* spreads primarily by vegetative growth of its roots. The root system can be extensive, growing horizontally as much as 20 feet in one season (Rogers 1928). Most patches spread at the rate of 3 to 6 feet per year (Amor and Harris 1975).

Cirsium arvense has two types of roots: horizontal and vertical. Horizontal roots produce numerous shoots, while vertical roots store water and nutrients in their many small branches.

Most *Cirsium arvense* roots are found directly below the aboveground shoots, with little extension beyond the border of a patch (Donald 1994). Apparently, the horizontal roots give rise to shoots frequently as they expand the range of a patch. Horizontal roots grow within 6 to 12 inches of the soil surface, and typically grow in a straight line for 2 to 3 feet, then bend down and grow vertically. Another horizontal root system is usually initiated at the downward bend (Rogers 1928). The horizontal roots are widest at the bend and can reach a maximum of 0.75 inch in diameter, although in sand, roots rarely exceed 0.3 inch in diameter (Rogers 1928).

Vertical roots can grow as deep as 22 feet (Rogers 1928), but most roots are in the upper 2 feet of soil (Haderlie et al. 1987). *Cirsium arvense* roots commonly reach a depth of 5 feet in 1-year-old plants and 6.5 feet in 2- to 10-year-old plants (Nadeau 1988). Root weight averages 40 ounces per square yard and decreases with depth, from 18 ounces per square yard in the top 1 foot to 12 ounces per square yard in the 1- to 2-foot depth, and 9 ounces per square yard in the 2- to 3-foot depth (Donald 1994).

Individual roots live up to 2 years (Rogers 1928). New root buds develop in autumn after the death of aerial shoots (McAllister 1982). Root bud development is highest under short days and moderate temperatures (autumn), and root bud elongation is greatest under long days and high temperatures (summer) (McAllister 1982).

Root growth and survival are affected by environmental factors, especially soil moisture, soil temperature, and substrate. Under high soil moisture, Canada thistle roots are susceptible to damping off (Bakker 1960). Root length increases in the top 1 foot of soil when growing season moisture is reduced, which increases drought tolerance in established plants (Lauridson et al. 1983). However, a dry winter can result in mortality due to desiccation of roots (Lauridson et al. 1983). In northern locales (Sweden), mild winters are linked to spread of *Cirsium arvense,* as growth begins earlier in the spring when more roots survive the winter (Gustavsson 1994). *Cirsium arvense* roots are cold-sensitive, injured when directly exposed to cold temperatures for 8 hours at –2 degrees Celsius, and dying after 8 hours at –6 degrees Celsius (Dexter 1937, Schimming and Messersmith 1988). Canada thistle roots usually survive subfreezing temperatures when insulated by soil, snow cover, and vegetative cover. Canada thistle roots also develop cold tolerance with increased exposure to the cold (Schimming and Messersmith 1988). It is suspected that deep roots (>1 foot below the soil surface) are more susceptible to freezing than shallow roots (Schimming and Messersmith 1988) because they do not develop cold tolerance. Root growth varies by substrate. The most extensive root growth occurs on moist clay, but growth is reduced on excessively wet soil and on droughty soil including sand, gravel, and hardpans (Rogers 1928).

Root carbohydrate reserves follow an annual cycle. Reserves are lowest early in June, just before flowering. Root reserves begin to increase early in fall as shoot growth declines (Hodgson 1968, Bakker 1960, Arny 1932, Welton, et al. 1929).

Shoots—Shoots begin to emerge when the average weekly temperature is 5 degrees Celsius, and emergence is highest when temperature is 8 degrees Celsius (Hodgson 1968). In Montana, shoots usually begin to emerge in the second week of May (Hodgson 1964) while in Nebraska they emerge beginning 22 March, and flowering begins about 1 June (Hoefer 1982). Growth is more vigorous under 25/15 degrees Celsius (day/night) regime than colder (15/5 °C) or warmer (30/22 °C) regimes, with 13 and 15 hours of light (Hoefer 1982). However, when the soil is warm (17 °C) and air temperature moderate (15/5 °C), as is common in autumn, Canada thistle grows vigorously (Hoefer 1982).

Primary shoots grow as rosettes for 2 to 4 weeks, then elongate (bolt) and develop flower buds some 10 weeks after emergence. Shoots elongate at the rate of

1 inch a day in late June, to an average of 4 feet (Hodgson 1964). Secondary shoots, produced from root buds, emerge throughout the summer. Thus, several growth stages may be simultaneously present.

Root buds are inhibited by the presence of the main shoot (both leaves and stem tissue), primarily because of competition for water between root buds and the main shoot (Hunter et al. 1985). When the main shoot is removed (e.g., as by mowing), the root buds are released and new shoots emerge rapidly, especially when humidity is high.

Most root buds are produced in the center of a patch (up to 80 per ft^2) near the soil surface (root bud density decreases with depth; Donald 1994). Each meter of root averages 12.8 to 24.4 root buds, each capable of forming a new shoot (Donald 1992). Nadeau and Vanden Born (1989) found an average of eight shoots are produced per meter of root.

Shoot density varies greatly, depending on substrate, moisture conditions, light availability, competition, and season, among other factors. Recorded shoot densities range from 4 per square yard (Hodgson 1964) to 276 per square yard (Donald 1994); averages of 7 to 84 per square yard are frequently reported (Donald 1992 and 1993b, Zimdahl and Foster 1993). Bakker (1960) found an average density of 5 shoots per square foot with 41 flower heads per shoot in open sites, and a density of 1.5 shoots per square foot with 18 flower heads per shoot in shaded areas. Shoot density varies across a patch and is usually densest near the center and lowest on the edges (Donald 1994).

Shoot density is positively correlated with rainfall during the previous growing season. It increased following a year of above-normal precipitation and decreased the year following a growing season drought (Donald and Prato 1992). In North Dakota shoot density approximately doubled between late summer and the following spring, from 2.4 shoots per square yard in August to 5.5 shoots per square yard the following June (Donald 1993). Shoot density and root growth are closely correlated. Areas with highest shoot density have the highest underlying root biomass and highest density of adventitious root buds, and also more deep roots (Donald 1994).

In established clones, shoot production increased with increased nitrogen (Nadeau 1988, Nadeau and Vanden Born 1990), indicating that *Cirsium arvense* infestations may be more severe on high-nitrogen soils. This may explain presence of *Cirsium arvense* in degraded wetlands or wetlands in which the water table is lowered. On the other hand, shoot production by young plants is stimulated more by favorable temperature and moisture regimes than by nitrogen levels (Nadeau 1988).

In Russia, Mikhailova and Tarasov (1989) determined that the majority of shoots in a clone were both mature and vegetative; less than 10 percent were either young or sexually reproductive.

Growth

Plants grow rapidly from seed, developing roots 4.5 feet deep at the end of the first growing season and flowering the second year (Rogers 1928). Seedlings first develop a branched primary root 2 to 4 inches deep, and then produce their first true leaves (Bakker 1960). Roots grow rapidly in young plants, up to 0.75 inch per day in the first 13 weeks (Nadeau 1988). A 4-month-old plant had a 40-inch root with 19 shoot buds (Detmers 1927). After just 18 weeks, plants averaged 36 feet of roots (Nadeau and Vanden Born 1989), 26 aboveground shoots, 154 underground shoots, and 364 feet of roots (diameter >1/50 inch). If these roots were cut into 4-inch-long pieces, each piece could have produced an additional 930 shoots.

Growth is strongly influenced by environmental factors. Seedlings require high light and low competition to survive (Moore 1975, Hodgson 1968, Bakker 1960). Thus, Canada thistle apparently has difficulty becoming established from seed in undisturbed areas. Amor and Harris (1974) reported no seedling establishment from seed artificially sown in pastures, whereas 7 to 13 percent of seeds sown on bare dirt emerged, and 78 to 93 percent of these seedlings became established. Seedlings grow rapidly under high humidity (90–100%), with a 50 percent increase in stem height and both shoot and root weight compared to seedlings growing at 50 percent humidity (Hunter, et al. 1985).

Drought may favor or disfavor *Cirsium arvense.* The plant's vigor decreases with drought conditions (Hansen 1918), especially in autumn (Boerboom and Wyse 1988a) although in Sweden, this species' long root system allows it to tolerate dry summers better than annual crops (Gustavsson 1994). Established plants develop drought tolerance by increasing root length in the top 1 foot of soil (Lauridson, et al. 1983). However, shoot density decreases the year following a growing season drought (Donald and Prato 1992).

Vegetative spread

Cirsium arvense spreads vegetatively through horizontal growth of the root system, which can extend 16 feet radially in one season (Bakker 1960). Individual clones can reach 115 feet in diameter (Donald 1994).

Cirsium arvense readily propagates from stem and root fragments, thus plowing or other soil disturbance can increase thistle densities (Nadeau and Vanden Born 1989). Small, root fragments (0.75 in) can survive and produce clones up to 9 feet across within 1 year (Rogers 1928). Hayden (1934) reported plants developing from root fragments as small as 0.2 inch and 95 percent establishment from 1-centimeter-long root fragments. Root fragments are able to produce new shoots independent of the presence of root buds (Nadeau 1988). Rogers (1928) stated that a 6-week-old root fragment could still regenerate a plant.

Partly buried stem fragments have much higher survival than fully buried fragments, as the cut stems remain photosynthetically active (Magnusson, et al. 1987). Regrowth from stem fragments is highest in mid-June (>70%) and lower thereafter (0–55%) (Magnusson et al. 1987).

Miscellaneous

Both roots and leaves may be mildly allelopathic. Extracts from roots and foliage reduced radicle growth, but did not inhibit germination, of several crop and weed species (Stachion and Zimdahl 1980).

American Indians quickly became familiar with *Cirsium arvense* and purportedly used an infusion of its roots for mouth diseases (Rousseay and Raymond 1945 [cited in Moore and Frankton 1974]). The Chippewa considered it to be "tonic, diuretic, and astringent" (Densmore 1928). Rogers (1928) indicated that young shoots and roots "can be used in the same ways as asparagus," and were eaten in Russia and by Native Americans. The nectar of *Cirsium arvense* flowers purportedly makes good honey (Rogers 1928).

Restoration potential

No studies on the recovery potential of *Cirsium arvense* infested areas were found, but recovery will be influenced by the control method employed. Areas treated with repeated disking, repeated mowing, or broadcast herbicide application usually have little or no native vegetation remaining. Areas treated with less aggressive techniques, such as prescribed fire, spot-applied herbicides, biocontrol agents, or infrequent mowing, generally retain most of the native community. Fire may be the least damaging treatment method because in many habitats it stimulates growth of native vegetation that subsequently competes with Canada thistle. Increasing the native component of the invaded community reduces the potential for *Cirsium arvense* reinvasion by decreasing bare soil (and opportunity for seedling establishment) and increasing competition (thereby reducing rate of vegetative invasion).

Combining biocontrol and prescribed fire or mowing may help control Canada thistle and promote restoration, but this is still in the experimental stage.

Management requirements

Cirsium arvense should be removed from high quality natural areas when it is first observed. The plant is tenacious and difficult to control once established. In lower quality areas, management effort should be influenced by the extent of invasion; greater effort is warranted in areas that have new and/or small invasions that are more likely to be eliminated or contained.

Management programs

Cirsium arvense management programs should be designed to kill established clones since the species spreads primarily by vegetative expansion of the root system. Prevention of seed production is a secondary consideration since spread by seeds is relatively rare. On the other hand, seedlings are the most susceptible growth stage (Bakker 1960). In areas that are susceptible to thistle invasion, but have not yet been invaded, management programs should be implemented to prevent the species from becoming established.

It is important to understand the biology of *Cirsium arvense* as control is greatly influenced by clonal structure (Donald 1994), growth stage (Tworkoski 1992), season of treatment, weather conditions, ecotype (Hodgson 1964), soil type, and control method(s) used. A single control method is rarely effective, and it is often necessary to use two or more methods at any given site (Donald 1992, Diamond 1993). In addition, treatments or combinations that are effective at one site may be ineffective at others (Frank and Tworkoski 1994).

It takes at least two growing seasons to determine whether a particular control method is effective. Several studies have recorded a temporary decline in *Cirsium arvense* in the first year after treatment, followed by a return to the pre-treatment conditions the second growing season (Zimdahl and Foster 1993).

The literature on Canada thistle control focuses on agricultural systems. Management in natural areas is more difficult because of the need to protect native species and communities. At this time, no control methods are suitable for widespread use in natural areas that eradicate, rather than reduce, Canada thistle.

Management strategies should be adjusted to reflect weather conditions. For example, drought stress reduces the effectiveness of most herbicides against *Cirsium arvense* (Haderlie et al. 1987), but increases the effectiveness of mechanical controls (Hansen 1918, Johnson 1912). Thus, mowing or burning would be preferred strategies under drought conditions.

Biological control

Overall, biocontrol currently provides little or no control of Canada thistle populations, although some agents weaken and kill individual plants. In North American, most potential biocontrol organisms are not adequately synchronized with the lifecycle of *Cirsium arvense* to induce high mortality. Management that delays *Cirsium arvense* maturation, such as mowing or burning, may help synchronize the susceptible thistle growth stage to the biocontrol agent lifecycle (Forsyth and Watson 1985a).

Cirsium arvense has few or no effective natural enemies in its native habitat, where it is also considered a severe agricultural weed (Peschken 1971). In all, more than 130 species, including diseases, birds, and more than 80 insects, attack Canada thistle (Maw 1976). At any one site in its native range, however, an average of 4.5 insect species attack *Cirsium arvense*, but they generally cause little damage as their densities are usually low and most species consume little plant material (Freese 1995). In North America, larvae of the native painted lady butterfly (*Vanessa cardui;* Lepidoptera) feed on Canada thistle and other related thistles and cause extensive defoliation within localized areas, but impact varies year to year because of their migration patterns (Story et al. 1985).

At least seven insect species have been intentionally or unintentionally released for Canada thistle control in North America, and a few of them cause conspicuous damage. The beetle *Cassida rubiginosa* was introduced accidentally in 1902 and defoliates plants (Maw 1976). Larvae of the intentionally introduced bio-control weevil *Ceutorhynchus litura* feed on stems of Canada thistle. The introduced stem-galling fly *Urophora cardui* attacks thistle shoots, but has little impact. Larvae of the fly *Orillia ruficauda* (Diptera) damage seed heads. The beetles *Altica carduorum* and *Lema cyanella* feed on Canada thistle's leaves. The seed weevil *Rhinocyllus conicus* was introduced to control musk thistle (*Carduus nutans*) and other related *Carduus* and *Cirsium* thistles and lays eggs in Canada thistle flowerheads. The weevil *Larinus planus* is a seedhead feeder, but it has had little impact on Canada thistle and attacks native thistles. Two pathogens have also been considered for use against Canada thistle. The rust *Puccinia punctiformis* and the fungus *Sclerotinia sclerotiorum* attack shoots and roots respectively. Of all these biocontrol organisms, *Orellia ruficauda* and *Puccinia punctiformis* appear to inflict the most significant damage (Maw 1976, Forsyth and Watson 1985a), but even this is probably not sufficient to control Canada thistle populations.

A combination of biocontrol agents, or of biocontrol agents and herbicides, may provide better control of Canada thistle than any single agent may. It has been suggested that at least three biocontrol organisms may be needed for effective Canada thistle control (Forsyth and Watson 1985a). In Western Canada, where *Cassida rubiginosa* and *Puccinia punctiformis* do not occur, *Cirsium arvense* is a much greater problem than in the eastern part of Canada, where these organisms are present. In Ontario, there appeared to be a synergistic relationship between infestation of thistle by *Ceutorhynchus litura* and infection by the rust *Puccinia punctiformis.* Weevils mined 87 percent of rust-infected thistles compared to 32 percent of the uninfected shoots (Peschken and Beecher 1970). Such an effect is not reported for sites in Western Canada (Peschken and Wilkinson 1981). Impacts of *Ceutorhynchus litura* are also enhanced when Canada thistle is infected with the fungal pathogen *Sclerotinia sclerotiorum. Ceutorhynchus* mining may have significant impacts after *Sclerotinia* infection under drought conditions, especially in the western Great Plains (Bourdot et al. 1995). Vegetative shoots were most susceptible to the disease (Bourdot et al. 1995), but the disease was not transferred to shoots that emerged after the initial infection. This disease is, therefore, most effective as a control method if applied after the majority of shoots have emerged. Disease development, however, requires high moisture conditions, which generally are less likely as the growing season progresses (Bourdot et al. 1995). Thus, timing of application is critical and varies between sites and years.

A combination of root- and shoot-feeding insects are needed for effective biological control, but no root feeders are known that cause substantial damage to *Cirsium arvense* (Ang, et al. 1995). The organisms tested for biological control were not simultaneously tested for tolerance to herbicides (Trumble and Kok 1982), but it appears that 2,4-D can be applied at low rates in conjunction with the rust *Puccinia punctiformis* to achieve better control than either treatment alone (Haggar, et al. 1986).

***Leaf-feeding Painted Lady Butterfly Vanessa cardui*—**Larvae of the native painted lady butterfly (*Vanessa cardui*) feed on *Cirsium arvense* and other *Cirsium* species and can defoliate and kill individual plants (Detmers 1927, Rees 1991). Painted lady typically occurs in Southern States, including California, and is itself infected by a virus that keeps its populations low. Every 8 to 11 years, populations explode and the butterflies migrate north where they can temporarily be effective biocontrol agents. Viral infection spreads rapidly in large painted lady butterfly populations, however, and within a year or two the butterfly populations drop again (Rees 1991).

Leaf-feeding Beetle Cassida rubiginosa*—Cassida rubiginosa* was accidentally introduced to the U.S. in 1902 (Barber 1916 [in Ang, et al. 1995]). This beetle causes severe defoliation of Canada thistle in Virginia and Maryland (Ang, et al. 1995), but only minimal damage in Quebec (Forsyth and Watson 1985a). Defoliation by *Cassida rubiginosa* is most effective at high insect density on young plants (Forsyth and Watson 1985a), but under field conditions this insect is not synchronized with young thistles and thus causes minimal damage. Ang et al. (1995) determined that *Cassida rubiginosa* significantly reduced thistle biomass and survival. At a density of 20 beetles per plant, over two-thirds of the thistles died by the end of the growing season. *Cassida* impact was substantially greater during drought conditions, and roots were devastated by attacks of 10 beetles per plant (Ang et al. 1995). However, damage by *Cassida rubiginosa* is rarely sufficient to reduce thistle growth in the field (Diamond 1993). *Cassida rubiginosa* larvae are themselves parasitized by fly, wasp, and beetle species (Tipping 1993). In addition, *Cassida* has low dispersal rates and rarely moves more than 6.5 feet from the release site within 26 days (Tipping 1993). Adults oviposit at the release point, regardless of thistle density (Tipping 1993). For effective control, beetles must be deposited on thistle rosettes at about 13-foot intervals, or at least in each patch within a site.

***Stem-mining weevil Ceutorhynchus litura*—** Between its initial introduction in North America in 1967 and 1985, the stem-mining weevil *Ceutorhynchus litura* became established in five Canadian provinces and Montana (Peschken and Wilkinson 1981, Story et al. 1985). *Ceutorhynchus litura* can reduce overwinter survival of *Cirsium arvense*, but thistle stands recover by shoot recruitment from unattacked plants (Rees 1990). Females feed on leaf tissues and lay eggs in feeding cavities. The developing larvae mine leaves and migrate inside stems to the root collars (Rees 1991). Unfortunately, *Ceutorhynchus litura* larvae mine the parenchyma tissue of the stem pith and do not damage vascular bundles, so water translocation is not affected (Peschken and Wilkinson 1981). While thistles usually survive the stem mining, the holes left by departing larvae provide entrance sites for other arthropods, nematodes, and disease organisms that cause high mortality of belowground shoots (Rees 1990). However, production of new shoots from underground roots the following spring offsets shoot mortality caused directly and indirectly by *Ceutorhynchus litura* (Rees 1990).

Seed Head Predator Orillia ruficauda*—Orillia ruficauda* is a small fly that deposits its eggs in *Cirsium arvense* flowerheads. Damage occurs when developing larvae eat the seeds in mid-summer (Detmers 1927). This may reduce seed production and seed dispersal (Forsyth and Watson 1985b). In one study 20 to 85 percent of seedheads were attacked, and 20 to 80 percent of seeds within each attacked seedhead were damaged. Forsyth and Watson (1985a) reported that *Orillia ruficauda* occurred in up to 70 percent of flowerheads and destroyed 22 percent (range 0–90%) of the seeds per head. Seed predation causes only limited suppression of *Cirsium arvense*, however, as the plant spreads primarily by vegetative means (Diamond 1993, Forsyth and Watson 1985a). While flies avoid laying eggs in male flowerheads and preferentially select female flowerheads, the developing larvae do not eat enough seeds in a flowerhead to affect either the individual seedhead or the population (Lalonde and Roitberg 1992b). Apparently, flies lay only a few eggs in any one flowerhead and avoid laying eggs in previously infested flower heads (Lalonde and Roitberg 1992a).

Other natural enemies—The Chrysomelid beetle *Altica carduorum* weakens Canada thistle by defoliating it and feeding on its flowerheads. It was first regarded as a promising control agent because of its specificity and continuous feeding habit, but has proven unsatisfactory because of its own susceptibility to predation (Peschken et al. 1970; Story et al. 1985; Schaber et al. 1975). *Cleonus piger* is a root-feeding weevil that can cause wilting and plant death, but plants usually regenerate from damaged vascular tissue (Forsyth and Watson 1985a). The leaf spot disease *Septoria cirsii* is host specific to *Cirsium arvense* and causes severe damage to *Cirsium arvense* plants in the field, inhibiting seed germination and root elongation and causing leaf chlorosis and necrosis (Hershenhorn et al. 1993). This disease has been proposed for consideration as a biological control organism.

Burning

Before providing assistance with a burn or burn recommendations, consult the NRCS burn policy in the general manual. *Cirsium arvense* response to fire varies from positive to negative, depending on season of burn, soil moisture, and location. Dormant season burning stimulates growth of native herbaceous species that compete with Canada thistle. Growing season fire damages native species as well as Canada thistle.

In a mesic grassland in Oregon, dormant season fire reduced Canada thistle flowerhead and seed production. Flowering plants had equal density in burned and unburned plots (7 to 8/ ft^2), but produced 50 percent fewer flowerheads in the burned plots (18 per shoot vs. 36 per shoot, respectively; Young 1986). Additionally, plants in the burned plots produced an average of 1.2 functional or seed-producing flowerheads per shoot, compared to 16.3 per shoot in the unburned plots. Dormant season burning (December or April) also stimulated production of numerous small Canada thistle shoots, resulting in higher density, but equal biomass (Young 1986).

In North Dakota, dormant season burning reduced the relative abundance of *Cirsium arvense* by stimulating growth of native vegetation (Carlson 1987). Growing season fires reduced thistle density but harmed native species (Smith 1985). Plots burned in mid-June had heavy seed production July through September, while plots burned mid-July to mid-August had numerous seedlings in August and September but they failed to survive the winter (Smith 1985).

In Alberta, Canada, spring burning in a marsh favored growth of native species and did not alter *Cirsium arvense* biomass. An August fire increased biomass and shoot density of *Cirsium arvense*, which averaged 2.2 ounces per square yard versus 6 grams per square yard, and 24 shoots per square yard versus 0.12 shoots per square foot, on burned and unburned plots, respectively (Thompson and Shay 1989). Seedling density also increased following the summer fire. In another wetland in Alberta, Canada, *Cirsium arvense* cover was not affected by fire (Hogenbirk and Wein 1991), but increased when the wetland area was subjected to drought.

Chemical control

Most studies of herbicide use are focused on reducing *Cirsium arvense* in agricultural areas and are not directly applicable to use in natural areas. For example, application of 2,4-D for Canada thistle control may be ineffective because it sets back the succession of natural communities, actually opening areas for thistle invasion. Other herbicides can similarly impact native vegetation.

The following factors should be considered when using herbicides against Canada thistle:

- Their effectiveness is contingent upon *Cirsium arvense* growth stage, environment (Tworkoski 1992), ecotype (Hodgson 1970), and genotype (Frank and Tworkoski 1994).

- Different ecotypes respond differently to the same herbicide, so what is effective at one locale, or on one clone, may not be effective in other locales or clones (Frank and Tworkoski 1994). The herbicides used at a site should vary to prevent clones tolerant to one herbicide from becoming dominant (Frank and Tworkoski 1994). When selecting an alternative herbicide, use one with a different mode of action (mechanism by which it kills plants) to minimize chances that plants are not tolerant to both herbicides.
- In many habitats *Cirsium arvense* goes dormant shortly after native species, so the window is limited to apply herbicides when native species will not be affected.
- Treat an entire clone because not all shoots and roots in a clone remain physically connected (Donald 1992).
- For all herbicides except 2,4-D, two or more applications give better control than a single application, regardless of seasonal sequence (spring-fall treatment gave equal control to fall-spring treatment; Zimdahl and Foster 1993, Donald 1993).
- *Cirsium arvense* is best treated with herbicides early in spring or in fall. Fall treatments are usually more effective than spring treatments (Haderlie et al. 1987, Darwent et al. 1994a, Lym and Zollinger 1995). Herbicide absorption is enhanced late in summer and in fall when plants are in the rosette stage (Hunter et al. 1990). The shoot-to-root translocation is greatest at that time (Darwent et al. 1994a). Hunter (1996) found that control is improved if thistles are cut in late July and the resprouts treated with glyphosate about 4 weeks later in late August (the 'August rosette stage'). Second best treatment time is at flower bud stage, when root reserves are lowest, particularly under droughty conditions (Haderlie et al. 1987). However, native species can be damaged by growing season herbicide application.

Canada thistle's deep, well-developed root system makes it resilient to most control methods including herbicides. However, *Cirsium arvense* undergoes several growth stages during the growing season, and during certain stages root carbohydrates are depleted. Root carbohydrate depletion is related to growth stage and is greatest when flowering occurs, but replenishment is related only to environmental conditions, and generally occurs in late summer and fall. Younger growth stages (spring) are most likely more susceptible to herbicide, but the root system is larger and more difficult to kill in spring before the flower stalk emerges. Older growth stages (fall) are somewhat less susceptible, but the root system is depleted and smaller, and assimilates are naturally moving from the leaf tissues to the root system (Tworkoski 1992). More assimilate (and hence herbicide) moves into the roots under short days and low temperatures (fall) than under long days and warm temperatures (summer; McAllister 1982).

Herbicide effect is enhanced when *Cirsium arvense* roots are weakened during the growing season by herbicide treatment, crop competition, or frequent mowing or tilling; and when new shoots are stimulated to grow. Suitable herbicides (e.g., glyphosate) should be applied to new growth when leaves are green (September or October). Avoid applying herbicide to old leaves (thick cuticle limits absorption) or to drought-stressed leaves.

Clopyralid (Stinger™ or Transline™), Clopyralid plus 2,4-D (Curtail™)—Clopyralid plus 2,4-D (sold under the tradename Curtail™) provides the best and most consistent control of Canada thistle in agricultural areas (Lym and Zollinger 1995), but may damage native forbs and shrubs. Clopyralid is a relatively selective post-emergence herbicide that kills certain broadleaf weeds and woody plants, but does little harm to others, such as members of the mustard family (*Brassicaceae*) or to grasses and other monocots. It is especially effective against members of the sunflower, buckwheat, and pea families (*Asteraceae*, *Polygonaceae*, and *Fabaceae*, respectively). The basis of this selectivity is not well understood for clopyralid or other auxin-type herbicides, such as 2,4-D or triclopyr (sold under the tradename Garlon™).

Clopyralid may have limited soil residual and is most effective on short (young) thistle shoots. Control was excellent on 2- to 6-inch-tall shoots, very good on 1-foot-tall flowering shoots, and poor on 2.5-foot-tall shoots (Donald 1992). Annual applications in early June at 2.5+9.8 ounces active ingredients per acre (clopyralid + 2,4-D) resulted in elimination or near elimination of all Canada thistle roots in the top 1.5 feet of soil after 2 to 4 years.

Fall application of Clopyralid delayed shoot emergence by 2 weeks and reduced shoot density the following summer (Donald 1993). The impact of clopyralid increased with increased application rate, and application of 72 ounces per acre had the greatest impact. One fall application with clopyralid at 48 ounces per acre prevented almost all *Cirsium arvense* shoot emergence the following spring.

Glyphosate (Roundup, Rodeo)—Glyphosate is a nonselective systemic herbicide that kills all vegetation green at the time of application. It has little or no soil residual. Glyphosate impacts *Cirsium arvense* by reducing the number of root buds and regrowth of secondary shoots, more than by reducing root biomass (Carlson and Donald 1988). No root bud regrowth occurred when glyphosate was applied at 24 ounces per acre (Carlson and Donald 1988). Translocation of glyphosate is significantly greater in plants at the bud to flowering stage than in younger plants (Sprankle, et al. 1975) and is greatest when plants are in the 'August rosette stage' (Darwent et al. 1994, Hunter 1995). The root is larger at the rosette stage, diluting the effect of glyphosate, but herbicide concentrations in the root are still up to three times higher at this time (Hunter 1995). This is because more herbicide is translocated when leaves are in vegetative (rosette and shoot-elongation) stage than in flowerbud or flowering stage (Hoefer 1982). In the laboratory, four times more glyphosate was translocated to the roots of rosettes than flowering plants (Hunter 1995).

Fall is the best season for applying glyphosate (Darwent et al. 1994, Lym and Zollinger 1995). For optimal results apply glyphosate under warm conditions before the first killing frost and when soil moisture is good, or after plants have adjusted to colder weather. Avoid treating thistles immediately before the first frost (Lym and Zollinger 1995). Plants treated with glyphosate one day before frost had much lower translocation of herbicide to the roots than plants in warm conditions, or plants hardened to cool air and soil temperatures (15 per 5 °C; Hoefer 1982).

Response of *Cirsium arvense* to glyphosate varies among clones (Frank and Tworkoski 1994, Darwent et al. 1994a). The majority of damage occurs after 3 days, but glyphosate continues to act on sensitive tissues for up to 45 days (Carlson and Donald 1988). Good soil moisture is important for glyphosate to be effective (Haderlie et al. 1987). Glyphosate impact was slightly reduced under severe drought conditions (Lauridson et al. 1983).

A low glyphosate concentration (2.5%) was more effective than higher concentrations (5%, 10%, and 30%), reducing shoot growth and regrowth 76 percent at the lower rate and having no effect at the higher rates (Boerboom and Wyse 1988b). At high concentrations glyphosate kills leaves so quickly that they are unable to translocate the herbicide to the roots before they die. Droplet size is also a factor as large droplets kill leaf tissue more than small droplets (Boerboom and Wyse 1988b). Lower levels of surfactant (MON 0818) are recommended, as glyphosate mixed with high MON 0818 concentrations may kill leaves rapidly (Boerboom and Wyse 1988b).

Haderlie et al. (1987) stated that glyphosate was most effective on fall regrowth, then at flower/bud stage, and least effective in spring when applied to 10-inch-tall plants. On the other hand, Devine (1981) found that although glyphosate translocation was slower under low temperatures, total uptake was not affected by growing conditions. After 5 days, 63 percent of the amount applied was absorbed and 22 percent exported to the roots regardless of temperature. Glyphosate was unevenly distributed in the root system and concentrated in fibrous roots and new shoot buds (Devine 1981). Between 1 and 2 percent of glyphosate was extruded by roots (pumped out into the surrounding soil) after 10 days (Devine 1981).

Cirsium arvense response to glyphosate differs between sites and/or clones. In Canada, a single application of glyphosate at 39 ounces per acre reduced thistle shoot density by more than 75 percent at two sites, while application at 155 ounces per acre was required to achieve the same level of control at a third site (Darwent et al. 1994a). Four consecutive annual applications at 39 ounces per acre reduced *Cirsium arvense* shoot density more than 98 percent at two sites, but at the third site four annual applications at 155 ounces per acre were required (Darwent et al. 1994a). Most reduction occurred after the first application at all sites.

One or two applications of glyphosate at 147 ounces per acre did not prevent *Cirsium arvense* shoot regrowth because enough roots remained to allow the plants to survive and resprout (Donald 1993). Fall treatment in two consecutive years decreased shoot density 94 percent the following fall, and root weight 77 percent (Carlson 1987). Five years after the treatments, however, thistle densities were the same in treated and untreated areas (Donald and Prato 1992).

Jaeger (pers. comm.) found application of Roundup to individual plants with a Walk-a-Wick™ applicator was difficult because the thistles were often below grass level. In 1985, park personnel in Minnesota began using a 4- to 5-gallon Solo backpack tank with the nozzle modified by a brass adjustment to apply a straight stream (not mist) at low pressure. Roundup at 3 to 4 percent was mixed with a purple agricultural dye and the mixture dribbled at the top of the stem and allowed to run down the stem. Use of the dye, which persisted as a marker of treated plants for up to a week, cut the time involved and the amount of herbicide used in half. Plants were treated in the pre-bud stage, and rounds were made weekly to assure treatment of plants that were missed earlier.

Chlorsulfuron—Chlorsulfuron is a post-emergent herbicide that primarily suppresses regrowth of Canada thistle and secondarily reduces the number of root buds and plant weight (Peterson 1983). Addition of growth regulators (chlorflurenol and dicamba) to chlorsulfuron enhanced control of *Cirsium arvense* in the greenhouse, but not under field conditions (Peterson 1983). Thistle density was reduced 2 to 5 years after spring application of chlorsulfuron (Donald and Prato 1992)

Not recommended

Picloram (Tordon™)—Picloram is a restricted use herbicide that may persist for up to 3 years in the soil and is not registered for use in California. It is relatively soluble and thus likely to be carried to the water table by percolating rain or irrigation water. Two to three annual fall applications of picloram at 24.2 ounces per acre gradually reduced *Cirsium arvense* density, and both one and three consecutive annual fall applications at 48.4 ounces per acre essentially eliminated *Cirsium arvense* (Donald 1993). Haderlie et al. (1987) found picloram was the most effective herbicide against Canada thistle, but it killed all broadleaved vegetation in treated areas. Picloram accumulates in shoot apices (Sharma and Vanden Born 1973 [cited in Donald 1990]) and is applied at flowerbud stage or to fall regrowth (Haderlie et al. 1987).

Dicamba (Banvel)—Dicamba has limited effectiveness on *Cirsium arvense* and persists for long periods in the soil, which makes it unacceptable for use in most natural areas. *Cirsium arvense* ecotypes vary in susceptibility to dicamba (Hodgson 1970, Saidak and Marriage 1976).

Metsulfuron (Ally)—Metsulfuron is ineffective against Canada thistle. Three consecutive fall applications did not reduce *Cirsium arvense* sufficiently (Donald 1993). Metsulfuron must be applied with another broadleaf herbicide, such as 2,4-D, to suppress Canada thistle (Lym and Zollinger 1995).

2,4-D—Canada thistle ecotypes varied greatly in their susceptibility to 2,4-D (Hunter and Smith 1972), and 2,4-D's impacts on treated plants were erratic (Donald 1990) and less effective than glyphosate or dicamba (Lym and Zollinger 1995). Three consecutive fall applications of 2,4-D did not reduce *Cirsium arvense* sufficiently (Donald 1993). Effectiveness of phenoxy herbicides, such as 2,4-D and MCPA, is greater when root carbohydrate reserves are low (Marriage 1981). In agricultural situations, a combination of 2,4-D with fertilization was effective under some circumstances. Hodgson (1968) found combining 2,4-D at 20 to 193 ounces per acre with 180 pounds per acre nitrogen and 605 pounds per acre phosphorus resulted in better thistle control and higher yields of spring wheat than either herbicide or fertilizer alone.

Five days after application 2,4-D was evenly distributed throughout the root system (Devine 1981). Of the applied 2,4-D, 3.1 percent is extruded from the roots (Devine 1981).

Bentazon (Basagran)—Bentazon is a post-emergent contact herbicide that can control topgrowth of Canada thistle when applied to plants roughly 8 inches tall in late spring and summer (Haderlie et al. 1987). At this time, however, native vegetation is susceptible to damage. Bentazon-induced chlorosis was evident in thistles emerging 10 months after treatment, indicating that Bentazon may be stored in roots over winter and transported back to the leaves in spring (Brewster and Stanger 1980).

Thistle density decreased more than 80 percent after single applications of Bentazon applied late May through late June (Brewster and Stanger 1980). Applications in early May treatment were less effective.

Repeated applications (two applications at 10– to 14-day intervals) of 190 ounces per acre provided better control than single applications at higher rates (294–580 oz/ac; Brewster and Stanger 1980). Split applications provided better control than a single application. A total of 72 ounces per acre resulted in 84 percent control with one application and 92 percent with two applications (Boerboom and Wyse 1988a). A total of 95 ounces per acre in one or two applications reduced *Cirsium arvense* by 40 percent and by 76 percent when applied in four applications (Boerboom and Wyse 1988a).

Grazing, dredging, and draining

Young plants are eaten by goats or sheep in the spring, but grazing is the least effective control method for Canada thistle (Rogers 1928). Cattle and horses avoid *Cirsium arvense* and browse on competing vegetation, which results in gradual dominance by *Cirsium arvense*. Heavy grazing breaks up sod, which permits seeding in of Canada thistle.

Data are not available on the effect of stocking rates or grazing intensities on Canada thistle. It seems likely that animal disturbance from conventional grazing encourages the spread of Canada thistle, as has been demonstrated for *C. lanceolatum*, *C. vulgare*, and *C. undulatum* (Tomarek and Albertson 1953, Ankle 1963, Hetzer and McGregor 1951).

Manipulation of water level and salinity

No information on the impacts of manipulating water levels on *Cirsium arvense* is available and little is available on the impact of manipulating soil salinity. Salt was one of the earliest chemicals used to kill *Cirsium arvense*. Applications of 180 to 640 pounds per acre used in the 1920's killed *Cirsium arvense* (Hodgson 1968), but the species is tolerant of lower salt concentrations. Seed germination is reduced, but not prevented by modest concentrations of NaCl. Wilson (1979) recorded germination rates of 83 percent without NaCl, 67 percent at 10,000 ppm NaCl, and 14 percent at 20,000 ppm NaCl (seawater is about 35,000 ppm salt).

Mowing, disking, and pulling

Mowing temporarily reduces aboveground biomass, but does not kill *Cirsium arvense* unless repeated at 7- to 28-day intervals for up to 4 years. This intensity of mowing is not recommended in natural areas where it would most likely damage native vegetation. Mowing just twice a year, in mid-June and September may reduce or contain Canada thistle. When mowing, cut high enough to leave more than 9 leaves per stem or more than 8 inches of bare stem tissue. Mature Canada thistle leaves and stems independently inhibit development of shoots from rootbuds. When the primary stem is removed, rootbuds are stimulated to produce new shoots that might otherwise be suppressed, especially under low humidity. Under high humidity root buds are stimulated to develop shoots regardless of presence of stem or leaves (Hunter et al. 1985). Cut plants also produce twice the length and weight of new shoots after just 7 days under high humidity (100%) than is produced under low humidity (50%; Hunter et al. 1985).

Early studies recommended mowing at frequent intervals to starve Canada thistle's root systems and remove it from farm fields and pastures (Cox 1913, Johnson 1912, Hansen 1918, Detmers 1929). Mowing monthly for a 4-year period eliminated practically all thistles (Welton et al. 1929), and mowing at 21-day intervals weakened roots and prevented seed production (Seely 1952). Hodgson (1968) found that mowing alfalfa fields twice annually, at Canada thistle's early-bud to pre-flowering stage (early to mid June in Montana) and early fall (September) reduced Canada thistle to 1 percent of its initial value in 4 years. Mowing two to three times a year can prevent seed set (Hansen 1913, Rogers 1928), but mowing once a year is ineffective (Donald 1990). To prevent production of viable seeds, stems must be mown before the flowers open when they have been open for only a few days. Stems with flowers that have been open 8 to 10 days can develop viable seeds (Derschied and Schultz 1960).

Tilling can reduce or eliminate *Cirsium arvense* if conducted repeatedly for several years. Tilling 3 to 4 inches deep every 21 days (6 cultivations over the growing season) reduced *Cirsium arvense* shoots by 98 percent in Montana (Hodgson 1958), and eradicated *Cirsium arvense* in Idaho (McKay et al. 1959). Ecotypes of *Cirsium arvense* differ somewhat in their response to repeated cultivation, but all were controlled by 10 cultivations over 1.5 growing seasons (Hodgson 1970). Tilling is not recommended in natural areas, however, because it would severely damage native vegetation and can sometimes spread Canada thistle across and between fields (Willard and Lewis 1939).

Tilling affects only the upper part of the root system, and in some cases as little as a quarter of Canada thistles roots are in the top 8 inches of soil reached by normal tillage. Most roots are 8 to 16 inches deep, and some reach to 6 feet deep (Nadeau and Vanden Born 1989). New shoots develop after tilling. Root fragments from a single young plant can produce more than 900 shoots when the roots are cut into 8-inch fragments (Nadeau and Vanden Born 1989), as typically occurs with disking. Deep tilling (4 to 8 inches below the soil surface) is more effective than shallow tilling (surface). Fewer shoots are present 40 days after deep tilling, and shoots cut at depth require more time to emerge than shoots cut near the surface (Darwent et al. 1994).

When tilling is discontinued early in August, new shoots do not produce flower stalks. Repeated tillage, however, kills *Cirsium arvense* by preventing shoot growth and thus depleting roots and their fragments of nutrient reserves (Donald 1990). Leaving large clods (2.07-inch diameter) minimizes seed germination, and leaving small clods (0.6-inch diameter) can stimulate germination of seedling that can be killed by retilling or treating with herbicide (Terpstra 1986). Seedlings should be disked or treated with herbicide within 2.5 weeks of thistle germination. After that, they will have developed roots that can survive disking and some herbicide treatments (Haderlie et al. 1987). Eight percent of seedlings (19 days old) with two true leaves resprouted when their tops were cut (Wilson 1979).

In Canada, the most successful control method for *Cirsium arvense* is the August rosette method, consisting of tilling until mid to late July, applying herbicide in mid August, and tilling again after 3 weeks (Alberta Agriculture 1993, Saskatchewan Agriculture and Food 1993, cited in Darwent et al. 1994b, Hunter 1996). Darwent et al. (1994b) recommend tilling *Cirsium arvense* patches until August 1, waiting 40 days for all shoots to emerge, and then applying glyphosate to the new shoots. Tilling until August 1 ensures that newly emerged shoots will remain as rosettes, as *Cirsium arvense* flowers only under long days. Waiting 40 days is necessary to obtain adequate shoot emergence and for shoots to grow large enough for effective glyphosate activity (Darwent et al. 1994b). This method is not practical in most natural areas unless thistles are mown or individually cut near the soil surface instead of tilled.

Reversing this procedure (applying herbicide and then tilling or disking) is ineffective regardless of herbicide type, season of herbicide application, or time between disking and herbicide treatment (Zimdahl and Foster 1993). Destroying shoots by disking releases dormant buds and may increase the total number of shoots (Zimdahl and Foster 1993). A minimum of 3 days between glyphosate and tilling is needed for glyphosate to damage the root system (Carlson and Donald 1988); waiting longer may further increase thistle mortality.

Disking in mid June is ineffective because cut stems readily develop new roots and establish new clones (Magnusson et al. 1987). Fewer cut stems survive when disking is conducted in mid September, and surviving stems do not develop adequate root systems to survive the winter (Magnusson et al. 1987). These roots are also more likely to be winterkilled, since disked fields accumulate less snow cover than undisked fields, and soil temperatures are lower (*Cirsium arvense* roots are injured at –2 degrees Celsius and killed at –8 degrees Celsius; Schimming and Messersmith 1988). Disking an all-male population may result in development of female plants.

Smothering

Mulching is impractical. Manure must be spread 5 feet thick and cover an area 16 to 20 feet in diameter. *Cirsium arvense* and plants that emerge at the mulch perimeter must be removed. Likewise, mulching with hay is ineffective because roots extend beyond the covered shoots (Willard and Lewis 1939). Mulching may actually enhance *Cirsium arvense* overwinter survival because mulch insulates cold-sensitive roots. However, covering Canada thistle with boards, sheet-metal, or tarpaper can kill the plants (Spence and Hulbert 1935).

Competition

Smother crops may be grown to choke and shade out undesirable species. To be effective against Canada thistle, the crop must come up first, grow rapidly during the early summer to shade out the thistle, and retain vigor until frost (Rogers 1928). These principles apply in the selection of smother crops on cultivated fields or haylands, but may also be applicable in the selection of native "smother" species in prairie restorations.

Smother crops are used in integrated pest management systems for Canada thistle on agricultural lands (Hodgson 1968), but the smother crops known to be effective are themselves invasive. Alfalfa (*Medicago sativa*) and especially sweet clover (*Melilotus alba* and/or *M. officinalis*) compete with and can reduce spread of *Cirsium arvense*, particularly when mowed as haycrops three times a year (Detmers 1927, Rogers 1928). However, these species are persistent and/or invasive in natural areas. Competition from timothy, orchardgrass, or redtop is ineffective (Detmers 1927).

Competition from tall fescue (*Festuca arundinacea*), in combination with the defoliating beetle *Cassida rubiginosa*, reduced *Cirsium arvense* density after 3 years, but not after 2 years (Ang 1993). Competition from tall fescue was more detrimental to *Cirsium arvense* than competition from crown vetch (*Coronilla varia*) (Ang et al. 1995), and damage increased when tall fescue was used in combination with *Cassida rubiginosa*.

Environmental stress

Control efforts may be more successful when *Cirsium arvense* is under environmental stress. The plant is drought and flood sensitive, and its roots are cold sensitive. Cutting or applying herbicide to shoots after a severe winter may add sufficient stress to kill plants.

Monitoring requirements

Monitor annually for presence of Canada thistle in a site. The best time to search is just before or during the blooming period, which varies from south to north, but corresponds with periods that have 14 to 18 hours of daylight (Linck and Kommendahl 1958, Hunter and Smith 1972). Once patches or individuals are located, remove or treat them before they flower and set seed. (Note that vegetative, and not sexual, reproduction is the primary method of expansion.) If Canada thistle is firmly established in a natural area, efforts should be made to eradicate, or at least to contain, the plant rather than simply monitor its spread.

Monitoring procedure

Walk through potential habitat—prairies, pastures, roadsides (any open herbaceous community). Abundance can be measured by recording the number of patches and the size of each patch along randomly located transects.

Bibliography

Amor, R.L., and R.V. Harris. 1974. Distribution and seed production of *Cirsium arvense* (L.) Scop. in Victoria, Australia. Weed Res. 14:317–323.

Ang., B.N. 1992. Multiple stresses by insect and plant competition on growth and productivity of Canada thistle (insect competition, *Cirsium arvense*, *Festuca arundinaceae*, *Coronilla varia*, *Cassida rubignosa*). Ph.D. thesis, Virginia Polytechnic Ins. and State Univ., 168 pp.

Ang, B.N., L.T. Kok, G.I. Holtzman, and D.D. Wolf. 1995. Canada thistle (*Cirsium arvense* (L.) Scop.) response to density of *Cassida rubignosa* (Coleoptera:Chrysomelidae) and plant competition. Biological Control 5:31–38.

Ankle, D.D. 1963. Vegetation and soil comparisons among three areas: mowed, relict, and moderately grazed. M.S. thesis, Fort Hays Kansas State Col., Fort Hays, KS, 45 pp.

Arny, A.C. 1932. Variations in the organic reserves in underground parts of five perennial weeds from late April to November. MN Agric. Exp. Sta. Tech. Bull. 84.

Bailiss, K.W., and I.M. Wilson. 1967. Growth hormones and the creeping thistle rust. Annals of Botany (London) 31:195–211.

Bakker, D. 1960. A comparative life-history study of *Cirsium arvense* (L.) Scop. and *Tussilago farfara* (L.), the most troublesome weeds in the newly reclaimed polders of the former Zuiderzee. *In* J. L. Harper, ed., The Biology of Weeds, Symp. No. 1, British Ecology Society, Blackwell Sci. Pub., Oxford, England, pp. 205–222.

Barber, H.S. 1916. A review of North American tortoise beetles. Proc. Entomological Soc. of Washington 18:113–127.

Boerboom, C.M., and D.L. Wyse. 1988a. Response of Canada thistle (*Cirsium arvense*) and birdsfoot trefoil (*Lotus corniculata*) to bentazon. Weed Sci. 36:250–253.

Boerboom, C.M., and D.L. Wyse. 1988b. Influence of glyphosate concentration on glyphosate absorption and translocation in Canada thistle (*Cirsium arvense*). Weed Sci. 36:291–295.

Boldt, P.F. 1981. Mechanical, cultural and chemical control of Canada thistle in horticultural crops. *In* Canada Thistle Symposium, Proc. N.C. Weed Control Conf. 36:179–180.

Bostock S.J., and R.A. Benton. 1983. Dry weight costs and establishment of seeds and vegetative propagules. Acta Oecologica/ Oecologia Plantarum 4(18):61—69.

Bourdot, G.W., I.C. Harvey, G.A. Hurrell, and D.J. Saville. 1995. Demographic and biomass production consequences of inundative treatment of *Cirsium arvense* with *Sclerotinia sclerotiorum*. Biocontrol Sci. and Tech. 5:11–25.

Brewster, B.D., and C.E. Stanger. 1980. Bentazon for Canada thistle (*Cirsium arvense*) control in peppermint (*Mentha piperita*). Weed Sci. 28:36–39.

Brosten, B.S., and D.C. Sands. 1986. Field trials of *Sclerotinia sclerotiorum* to control Canada thistle (*Cirsium arvense*). Weed Sci. 34:377–380.

Carlson, S.J. 1987. Glyphosate efficacy and induced biological responses in Canada thistle (*Cirsium arvense*) shoots, roots, and adventitious root buds; a washer for separating thickened roots from soil. Ph.D. thesis, North Dakota State Univ. of Agric. and Applied Sci., 127 pp.

Carlson, S.J., and W.W. Donald. 1988. Glyphosate effects on Canada thistle (*Cirsium arvense*) roots, root buds, and shoots. Weed Research 28:37–45.

Cockayne, A.H. 1915. California thistle rust. J. Agric. 12:300–302.

Cox, H.R. 1913. Controlling Canada thistles. U.S. Dep. Agric., Farmers Bull. 545, 14 pp.

Darwent, A.L., K.J. Kirkland, M.N. Baig, and L.P. Lefkovitch. 1994a. Preharvest applications of glyphosate for Canada thistle (*Cirsium arvense*) control. Weed Tech. 8:477–482.

Darwent, A.L., L. Townley-Smith, and L.P. Lefkovitch. 1994b. Comparison of time and depth of last tillage on the growth of Canada thistle (*Cirsium arvense*) in summer fallow and its response to glyphosate. Canadian J. Plant Sci. 74:867-873.

Densmore, F. 1928. Uses of plants by the Chippewa Indians. *In* Forty-fourth Annual Report Bur. Amer. Ethnology to the Sec., Smithsonian Inst., 1926-1927, originally published by U.S. Gov. Print. Of., Washington, 1928, and reprinted 1974 as How Indians use wild plants for food, medicine, and crafts, by Dover Pub., NY, pp. 275–397.

Derschied, L.A., and R.E. Schultz. 1960. Achene development of Canada thistle and perennial sow thistle. Weeds 8:59-62.

Detmers, F. 1927. Canada thistle (*Cirsium arvense* Tourn.), field thistle, creeping thistle. OH Agric. Exp. Sta. Bul. 414, 45 pp.

Devine, M.D. 1981. Glyphosate uptake, translocation and distribution in quackgrass (*Agropyron repens* (L.) Beauv.) and Canada thistle (*Cirsium arvense* (L.) Scop.). Ph.D. thesis, Univ. Guelph (Canada).

Dewey, S.A. 1991. Weedy thistles of the Western United States. *In* James, L.F., J.O. Evans, M.H. Ralphs, and R.D. Child, ed., Noxious range weeds, Westview Press, Boulder, CO, pp. 249–253.

Dexter, S.T. 1937. The winterhardiness of weeds. J. Amer. Soc. of Agron. 29:507—528.

Diamond, J. 1993. Integrated control of *Cirsium arvense* (L.) Scop. in pastures. Ph.D. thesis, McGill Univ. (Canada), 186 pp.

Dikova, B. 1989. Wild-growing hosts of the cucumber mosaic virus (Abstract). Rasteniev"dni Nauki 26(7):57–64.

Donald, W.W. 1990. Management and control of Canada thistle (*Cirsium arvense*). Reviews of Weed Sci. 5:193–250.

Donald, W.W. 1992. Herbicidal control of *Cirsium arvense* (L.) Scop. roots and shoots in no-till spring wheat (*Triticum aestivum* L). Weed Res. 32:259–266.

Donald, W.W. 1993a. Retreatment with fall-applied herbicides for Canada thistle (*Cirsium arvense*) control. Weed Sci. 41:434–440.

Donald, W.W. 1993b. Root versus shoot measurements to evaluate recovery of Canada thistle (*Cirsium arvense*) after several years of control treatments. Canadian J. Plant Sci. 73:369–373.

Donald, W.W. 1994. Geostatistics for mapping weeds, with a Canada thistle (*Cirsium arvense*) patch as a case study. Weed Sci. 42:648–657.

Donald, W.W., and T. Prato. 1992. Effectiveness and economics of repeated sequences of herbicides for Canada thistle (*Cirsium arvense*) control in reduced-till spring wheat (*Triticum aestivum*). Canadian J. Plant Sci. 72:599–618.

Ellis, W.N., and A.C. Ellis-Adam. 1992. Flower visits to *Cirsium* and *Carduus* (abstract). Entomologische Berichten (Amsterdam) 52:137–140.

Forsyth, S.F. 1983. Stress physiology and biological weed control: a case study with Canada thistle (*Cirsium arvense* (L.) Scop.). Ph.D. thesis. McGill Univ. (Canada).

Forsyth, S.F., and A.K. Watson. 1985a. Stress inflicted by organisms on Canada thistle. *In* Delfosse, E.S., ed., Proc. Fourth Intl. Symp. on Biol. Control of Weeds, Aug. 1984, Vancouver, Canada, pp. 425–431.

Forsyth, S.F., and A.K. Watson. 1985b. Predispersal seed predation of Canada thistle (*Cirsium arvense*). Canadian Entomologist 117:1075–1082.

Frank, J.R., and T.J. Tworkoski. 1994. Response of Canada thistle (*Cirsium arvense*) and leafy spurge (*Euphorbia esula*) clones to chlorsulfuron, clopyralid, and glyphosate. Weed Tech. 8:565–571.

Frantzen, J. 1994a. An epidemiological study of *Puccinia punctiformis* (Str.) Rohl as a steppingstone to the biological control of *Cirsium arvense* (L.) Scop. New Phytologist 127:147–154.

Frantzen, J. 1994b. The role of clonal growth in the pathosystem *Cirsium arvense-Puccinia punctiformis*. Canadian J. Botany 72:832–836.

Freese, G. 1995. The insect complexes associated with the stems of seven thistle species. Entomologia Generalis 19:191–207.

French, R.C., S.E. Nester, and R.G. Binder. 1994. Volatiles from germinating Canada thistle seed and root cuttings that stimulate germination of teliospores of the Canada thistle rust fungus, *Puccinia punctiformis*. J. Agric. and Food Chem. 42:2937–2941.

Gustavsson, A.M.D. 1994. Canada thistle occurrence and biology (abstract). Vaxtskyddsnotiser 58(3):79–84.

Haderlie, L.C., S. Dewey, and D. Kidder. 1987. Canada thistle biology and control. Bul. No. 666, Univ. Idaho Coop. Ext. Serv., 7 pp.

Haggar, R.J., A.K. Oswald, and W.G. Richardson. 1986. A review of the impact and control of creeping thistle (*Cirsium arvense* L.) in grassland. Crop Protection 5:73–76.

Hansen, A.A. 1918. Canada thistle and methods of eradication. USDA. Farmers Bul. 1002, 15 pp.

Hayden, A. 1934. Distribution and reproduction of Canada thistle in Iowa. Amer. J. Botany 21:355–373.

Hershenhorn, J.M. Vurro, M.C. Zonno, A. Stierle, and G. Strobel. 1993. *Septoria cirsii*, a potential biocontrol agent of Canada thistle and its phytotoxin-beta nitropropionic acid. Plant Sci. (Limerick) 94:227–234.

Hetzer, W.A., and R.L. McGregor. 1951. An ecological study of the prairie and pasture lands in Douglas and Franklin Counties, Kansas. Kansas Acad. Sci. Trans. 54:356–369.

Hodgson, J.M. 1964. Variations in ecotypes of Canada thistle. Weeds 12:167–171.

Hodgson, J.M. 1968. The nature, ecology, and control of Canada thistle. USDA Tech. Bul. 1386, 32 pp.

Hodgson, J.M. 1970. The response of Canada thistle ecotypes to 2,4-D, amitrole, and intensive cultivation. Weed Sci. 18:253–255.

Hoefer, R.H. 1981. Canada thistle (*Cirsium arvense*) root bud initiation, biology, and translocation of carbon-14 labeled glyphosate as influenced by nitrogen, temperature, photoperiod, and growth stage. Ph.D. thesis, Univ. Nebraska-Lincoln, 82 pp.

Hogenbirk, J.C., and R.W. Wein. 1991. Fire and drought experiments in northern wetlands: a climate change analogue. Canadian J. Botany 69:1991–1997.

Hope, A. 1927. The dissemination of weed seeds by irrigation water in Alberta. Sci. Agric. 7:268–276.

Hunter, J.H. 1995. Effect of bud vs. rosette growth stage on translocation of ^{14}C-Glyphosate in Canada thistle (*Cirsium arvense*). Weed Sci. 43:347–351.

Hunter, J.H. 1996. Control of Canada thistle (*Cirsium arvense*) with glyphosate applied at the bud vs. rosette stage. Weed Sci. 44:934–938.

Hunter, J.H., A.I. Hsiao, and G.I. McIntyre. 1985. Some effects of humidity on the growth and development of *Cirsium arvense*. Botanical Gazette 146:483–488.

Hunter, J.H., and L.W. Smith. 1972. Environmental and herbicide effects on Canada thistle ecotypes (*Cirsium arvense*). Weed Sci. 20:163–167.

Jansson, A. 1991. Distribution and dispersal of *Urophora cardui* (Diptera, Tephritidae) in Finland in 1985–1991. Entomologica Fennica 2:211–216.

Jessep, C.T. 1989. *Cirsium arvense* (L.) scopoli, Californian thistle (Asteraceae). *In* Tech. Common, ch. 60, Commonwealth Inst. Biol. Control (Wellingford, Oxon, UK), pp. 343–345.

Johnson, A.G. 1912. Canada thistle and its eradication. Purdue Univ. Agric. Exp. Sta., Cir. No. 32, Lafayette IN, 12 pp.

Kay, Q.O.N. 1985. Hermaphrodites and subhermaphrodites in a reputedly dioecious plant, *Cirsium arvense* (L.) Scop. New Phytologist 100:457–472.

Lauridson, T.C., R.G. Wilson, and L.C. Haderlie. 1980. Effect of drought stress on Canada thistle control. Proc. NC Weed Control Conf. 35:17.

Lauridson, T.C., R.G. Wilson, and L.C. Haderlie. 1983. Effect of moisture stress on Canada thistle (*Cirsium arvense*) control. Weed Sci. 31:674–680.

Lalonde, R.G., and B.D. Roitberg. 1989. Resource limitation and offspring size and number trade-offs in *Cirsium arvense* (Asteraceae). Amer. J. Botany 76:1107–1111.

Lalonde, R.G., and B.D. Roitberg. 1992a. Host selection behavior of a thistle-feeding fly: choices and consequences. Oecologia 90:534–539.

Lalonde, R.G., and B.D. Roitberg. 1992b. Field studies of seed predation in an introduced weedy thistle. Oikos 65:363–370.

Lalonde, R.G., and B.D. Roitberg. 1994. Mating system, life-history, and reproduction in Canada thistle (*Cirsium arvense*; Asteraceae). Amer. J. Botany 81:21–28.

Lhotska, M., and M. Holub. 1989. Influence of bovine digestive tract on germination of diaspores of selected plant species. Biologicia (Bratislava) 44:433–440.

Linck, A.J., and T. Kommendahl. 1958. Canada thistle-spotlight on a troublesome weed. Minnesota Farm and Home Sci. 15:21–22.

Littlefield, J.L. 1986. Host plant suitability of various clones of Canada thistle to gall induction and host selection by *Urophora cardui* (L.) (Diptera: Tephritidae), an introduced biological control agent. Ph.D. thesis, Univ. Wyoming, 112 pp.

Lloyd, D., and A.J. Myall. 1976. Sexual dimorphism in *Cirsium arvense* (L.) Scop. Annals of Botany 40:115–123.

Lym, R.G., and R. Zollinger. 1995. Perennial and biennial thistle control. North Dakota State Univ. Ext. Serv. Pub. W–799, Fargo ND.

Madsen, S.B. 1962. Germination of buried and dry stored seeds, III, 1934–1960. Proc. Intl. Seed Testing Assoc. 27:920–928.

Magnusson, M.U., D.L. Wyse, and J.M. Spitzmueller. 1987. Canada thistle (*Cirsium arvense*) propagation from stem sections. Weed Sci. 35:637–639.

Malicki, L., and C. Berbeciowa. 1986. Uptake of more important mineral components by commonfield weeds on loess soil. Acta Agrobotanica 39:129–142.

Marriage, P.B. 1981. Response of Canada thistle to herbicides. *In* Canada thistle Symp., Proc. NC Weed Control Conf. 36:162–167.

Maw, M.G. 1976. An annotated list of insects associated with Canada thistle (*Cirsium arvense*) in Canada. Canadian Entomologist 108:235–244.

McAllister, R.S. 1982. Influence of environmental factors on root bud growth and development, and on assimilate and glyphosate translocation in Canada thistle (*Cirsium arvense*). Ph.D. thesis, Univ. Nebraska-Lincoln, 116 pp.

McKay, H.C., P. Ames, J.M. Hodgson, and L.C. Erickson. 1959. Control Canada thistle for greater profits. Idaho Agric. Exp. Sta. Bul. 321, 14 pp.

Mikhailova, N.F., and A.V. Tarasov. 1989. The character of *Cirsium arvense* thickets. (Abstract). Botanicheskii Zhurnal (Leningrad) 74:509–514.

Moore, R.J. 1975. The biology of Canadian weeds. 13:*Cirsium arvense* (L.) Scop., Canadian J. Plant Sci. 55:1033–1048.

Moore, R.J., and C. Frankton. 1974. The thistles of Canada. Research Branch Canada Dep. Agricule. Mono. No. 10., Ottawa, Canada, 111 pp.

Nadeau, L.B. 1988. The root system of Canada thistle (*Cirsium arvense* (L.) Scop.): nitrogen effects on root bud dormancy. Ph.D. thesis, Univ. Alberta (Canada).

Nadeau, L.B., and W.H. Vanden Born. 1989. The root system of Canada thistle. Canadian J. Plant Sci. 69:1199–1206.

Osoki, K.L., P.K. Fay, B.K. Salley, E.L. Sharp, and D.C. Sands. 1979. Use of Canada thistle rust as a biological control agent. Proc. West. Weed Sci. Soc. 32:61.

Peschken, D.P. 1971. *Cirsium arvense* (L.) Scop., Canada thistle (Compositae). Commonwealth Inst. Biol. Control, Tech. Comm. 4:79–83.

Peschken, D.P., H.A. Friesen, N.V. Tonks, and F.L. Barnham. 1970. Releases of Altica carduorum (Chrysomelidae, Coleoptera) against the weed Canada thistle (*Cirsium arvense*) in Canada. Canadian Entomologist 102:264–271.

Peschken, D.P., and D.P. Wilkinson. 1981. Biocontrol of Canada thistle *Cirsium arvense:* releases and effectiveness of *Ceutorhynchus litura* (Coleoptera:Curculionidae) in Canada. Canadian Entomologist 107:1101–1110.

Peschken, D.P., and J.L. Derby. 1992. Effect of *Urophora cardui* (l.) (Diptera:Tephritidae) and *Ceutorhynchus itura* (F.) (Coleoptera:Curculionidae) on the weed Canada thistle, *Cirsium arvense* (L.) Scop. Canadian Entomologist 124:145–150.

Peterson, P.J. Absorption, translocation, and metabolism of chlorsulfuron and the effects of herbicide-growth regulator combinations on Canada thistle (*Cirsium arvense*) control. Ph.D. thesis, Univ. Nebraska-Lincoln, 75 pp.

Rees, N.E. 1990. Establishment, dispersal, and influence of *Ceutorhynchus litura* on Canada thistle (*Cirsium arvense*) in the Gallatin Valley of Montana. Weed Sci. 38:198–200.

Rees, N.E. 1991. Biological control of thistles. *In* James, L.F., J.O. Evans, M.H. Ralphs, and R.D. Child, eds. Noxious range weeds, Westview Press, Boulder, CO, pp. 264–273.

Roberts, H.A., and R.J. Chancellor. 1979. Periodicity of seedling emergence and achene survival in some species of *Carduus*, *Cirsium* and *Onopordum*. J. Applied Ecol. 16:641–647.

Rogers, C.F. 1928. Canada thistle and Russian knapweed and their control. Colorado Agric. Col. Colorado Exp. Sta., Fort Collins, Bul. 348, 44 pp.

Rotheray, G.E. 1986. Effect of moisture on the emergence of Urophora cardui (L) (Diptera: Tephritidae) from its gall on *Cirsium arvense* (L). Entomological Gazette 37(1):41–44.

Saidak, W.J., and P.B. Marriage. 1976. Response of Canada thistle varieties to amitrole and glyphosate. Canadian J. Plant Sci. 56:211–214.

Schaber, B.D., E.V. Balsbaugh, and B.H. Kantack. 1975. Biology of the flea beetle, *Altica carduorum* (Coleoptera:Chrysomelidae) on Canada thistle (*Cirsium arvense*) in South Dakota. Entomophaga 20:325–335.

Schimming, W.K., and C.G. Messersmith. 1988. Freezing resistance of overwintering buds of four perennial weeds. Weed Sci. 36:568–573.

Seely, C.I. 1952. Controlling perennial weeds with tillage. Idaho Agric. Exp. Sta. Bul. 288, 43 pp.

Smith, K.A. 1985. Canada thistle response to prescribed burning (North Dakota). Rest. and Manag. Notes 3:87.

Spence, H.L., and H.W. Hurlbert. 1935. Idaho perennial weeds. Their description and control. Univ. Idaho Col. Agric. Ext. Bul. 98, 30 pp.

Sprankle, O., W.F. Meggitt, and D. Penner. 1975. Absorption, action, and translocation of radioactive glyphosate. Weed Sci. 23:235–240.

Stachion, W.J., and R.L. Zimdahl. 1980. Allelopathic activity of Canada thistle (*Cirsium arvense*) in Colorado. Weed Sci. 28:83–86.

Story, J.M., H. DeSmet-Moens, and W.L. Morrill. 1985. Phytophagous insects associated with Canada thistle *Cirsium arvense* in southern Montana. J. Kansas Entomological Soc. 58:472–478.

Terpstra, R. 1986. Behavior of weed seed in soil clods. Weed Sci. 34:889–895.

Thomas, R.F., T.J. Tworkoski, R.C. French, and G.R. Leather. 1994. *Puccinia punctiformis* affects growth and reproduction of Canada thistle (*Cirsium arvense*). Weed Tech. 8:488–493.

Thompson, D.J., and J.M. Shay. 1989. First-year response of a *Phragmites* marsh community to seasonal burning. Canadian J. Botany 67:1448–1455.

Tipping, P.W. 1993. Field studies with *Cassida rubignosa* (Coleoptera:Chrysomelidae) in Canada thistle. Environmental Entomology 22:1402–1407.

Toole, E.H., and E. Brown. 1946. Final results of the Duval buried seed experiment. J. Agric. Research 72:201–210.

Tomarek, G.W., and F. W. Albertson. 1953. Some effects of different intensities of grazing on mixed prairies near Hays, Kansas. J. Range Manag. 6:299–306.

Trumble, J.T., and L.T. Kok. 1982. Integrated pest management techniques for the thistle suppression in pastures of North America. Weed Res. 22:345–359.

Turner, S., P.K. Fay, E.L. Sharp, B. Sallee, and D. Sands. 1980. The susceptibility of Canada thistle (*Cirsium arvense*) ecotypes to a rust pathogen (*Puccinia obtegans*). Proc. Western Weed Sci. Soc. 33:110–111.

Tworkoski, J.J., and J.P. Sterrett. 1985. Canada thistle control with combinations of growth regulators and glyphosate or triclopyr. Proc. Northeastern Weed Sci. Soc. 39:98.

Tworkoski, T. 1992. Developmental and environmental effects on assimilate partitioning in Canada thistle (*Cirsium arvense*). Weed Sci. 40:79–85.

Van Bruggan, T. 1976. The vascular plants of South Dakota. Iowa State Univ. Press, Ames. 538 pp.

Welton, F.A., V.H. Morris, and A.J. Hartzler. 1929. Organic food reserves in relation to the eradication of Canada thistle. Ohio Agric. Exp. Sta. Bul. 441.

White, D.J., E. Haber, and C. Keddy. 1993. Invasive plants of natural habitats in Canada. Canadian Wildlife Serv., Canadian Museum Nature, Ottawa, Ontario, 121 pp.

Willard, C.J., and R.D. Lewis. 1939. Eradicating Canada thistle. Ohio State Univ. Agric. Col. Ext. Serv. 146, 8 pp.

Wilson, R.G. 1979. Germination and seedling development of Canada thistle (*Cirsium arvense*). Weed Sci. 27:146–151.

Young. R.C. 1986. Fire ecology and management of plant communities of Malheur National Wildlife Refuge, southeastern Oregon. Ph.D. thesis, Oregon State Univ., 124 pp.

Youssef, N.N., and E.W. Evans. 1994. Exploitation of Canada thistle by the weevil *Rhinocyllus conicus* (Coleoptera:Curculionidae) in northern Utah. Environ. Entomology 23:1013–1019.

Zimdahl, R.L., and G. Foster. 1993. Canada thistle (*Cirsium arvense*) control with disking and herbicides. Weed Tech. 7:146–149.

III.J.2.b Management and control of leafy spurge (*Euphorbia esula* L.)(EUES)

(David D. Biesboer, Ph.D., The Nature Conservancy, Arlington, Virginia, author of original abstract 1996; Nancy Eckardt, The Nature Conservancy, Arlington, Virginia, revised 2000; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001. Photographs by Cami Dixon, NRCS, Jamestown, North Dakota; State Distribution Map, NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

This abstract provides information on the biology and control of invasive plant species in wetlands.

To the user

Element Stewardship Abstracts (ESAs) are prepared to provide The Nature Conservancy's Stewardship staff and other land managers with management-related information on those species and communities that are most important to protect, or most important to control. The abstracts organize and summarize data from numerous sources including literature and researchers and managers actively working with the species or community.

We hope, by providing this abstract free of charge, to encourage users to contribute their information to the abstract. This sharing of information will benefit all land managers by ensuring the availability of an abstract that contains up-to-date information on management techniques and knowledgeable contacts. Contributors of information will be acknowledged within the abstract and receive updated editions.

For ease of update and retrievability, the abstracts are stored on computer at the national office of The Nature Conservancy. This abstract is a compilation of available information and is not an endorsement of particular practices or products.

Please do not remove this cover statement from the attached abstract.

Content

The Nature Conservancy
Element Stewardship Abstract

Leafy Spurge (*Euphorbia esula*)

Identifiers

Common name: Leafy spurge Global rank: G5

General description: *Euphorbia esula* is a perennial plant ranging from 6 to 36 inches in height.

Stewardship summary

Monitoring of areas with known or potential *Euphobia esula* infestations is critical; adequate control is possible if management procedures are implemented in the early stages of infestation. Eradication of spurge is rarely achieved, but infestations can be reduced to manageable levels with the use of herbicides. Picloram is the most effective, and 2 pounds per acre applied in the spring and again in fall provides 85 to 90 percent control for several years. A less expensive and also effective method is to mix picloram at 0.25 pound per acre with 2,4-D at 1 pound per acre. This mixture applied once a year in the spring gives 90 to 95 percent control after about 5 years.

Whatever the treatment, it is important to realize that spurge cannot be controlled with a single herbicide treatment. Continuous surveillance and reapplication of the herbicide as shoot control decreases must continue for at least 10 years, and probably a good deal longer. For example, management at Devil's Tower National Monument has been spraying on an annual basis for about 20 years and has significantly reduced, but not eradicated spurge populations.

Prescribed burning in conjunction with herbicide application can provide excellent control of leafy spurge in open areas. Results are apparently good whether burning is followed by spraying or vice versa, but as with other methods, repeated treatments are necessary over at least a 5 to 10 year period.

Control of spurge in wooded or riparian zones can be extremely difficult since picloram is not labeled for use in these areas. Glyphosate and 2,4-D are commonly employed under trees with mixed results.

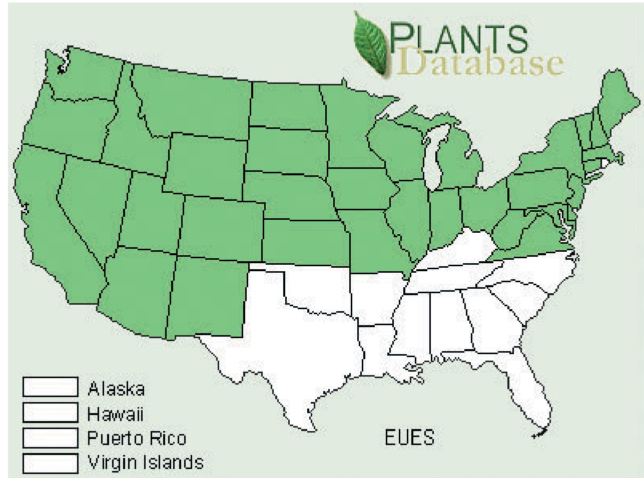
Biological control is being actively researched at many locations. Since the 1960's several insects have been released in certain locations, most notably the spurge hawkmoth, *Hyles euphorbiae.* Biocontrol agents alone have not so far been effective in controlling spurge populations, but may become valuable if several different insects can be successfully used together or in conjunction with other control methods. Research should focus on a highly integrated approach to spurge management with the goal of reducing the amount of herbicides needed for adequate control.

Natural history

Range

Euphorbia esula and its closely related taxa are native to central and Eastern Europe with extensions into Western Europe and temperate Asia. It is now found worldwide with the exception of Australia. It was most likely introduced into North America via Minnesota with shiploads of oats (Batho 1932).

Euphorbia esula is presently a major economic concern in the Northwestern and Northcentral States of the United States and in the adjacent prairie regions of the provinces of Canada. States with the greatest infestations include Colorado, Idaho, Minnesota, Montana, Nebraska, North Dakota, Oregon, South Dakota, Wisconsin, and Wyoming.



Habitat

Euphorbia esula occurs primarily in untilled, noncropland habitats, which include disturbed and undisturbed sites, such as abandoned cropland, pastures, rangelands, woodlands, prairies, roadsides, and wastelands. It is tolerant of a wide range of habitats and may occur in rich, damp soil, such as on streambanks or in extremely nutrient poor, dry soil typified by the rangelands of the West. It is most aggressive in semiarid situations where competition from associated species is less intense. For this reason, infestations generally occur and spread rapidly on dry hillsides, dry prairies, or rangelands. The plants tend to occur on all soils, but tend to grow most rapidly in coarse-textured soils (Selleck et al. 1962).

Ecology

Phenology—*Euphorbia esula* is one of the first plants to emerge in the spring. It emerges in early April in North Dakota, during March in Iowa and Wisconsin, and late April in Saskatchewan (Hanson and Rudd 1933, Bakke 1936, Selleck et al. 1962). Stem elongation is very rapid as daily temperatures increase from May through June. Seedlings may emerge when temperatures are near freezing. Seedlings appear deep red or purplish because of anothcyanin production in the hypocotyl. As the growing season progresses, some seedlings appear to dry up and die, but their underground parts will persist and produce adventitious buds, especially near the hypocotylar end of the shoot. The main seedling shoot usually does not survive and flower because of the rapid development of adventitious organs. It is replaced by an adventitious shoot that matures into the flowering shoot.

Infloresences form on the main axis from May to the end of July with flowering and seed development again occurring for a short time in the fall, usually from auxiliary branches. Seed development and maturation continue for 4 to 6 weeks after the appearance of the last flowers with seed dispersal occurring into early August. The plant usually ceases to grow during the hottest and driest weeks of July and August. Stems from seedling or root buds generally do not flower the first year. During senescence in the fall, the plants turn a pleasant golden-yellow or reddish-yellow before the leaves fall from the plant. The naked stem axis is woody enough to persist from summer to summer, and remnants of it can be seen at the base of newly emerged shoots. As light becomes limiting, plants fail to flower, decrease in density, and increase in height. As patches develop, density reaches over 240 shoots per square yard in light soil and up to 2,400 per square yard in heavy soil. In heavy soil about 60 percent of the shoots are produced from seed, whereas in light soil density is maintained and increased mainly by vegetative reproduction (Selleck 1959).

Maintenance—Leafy spurge, once established, will spread very rapidly, crowding out and shading desirable species. It emerges earlier in spring than most other species and shows allelopathy toward associated species as evidenced by bare ground and lack of other forbs in dense patches of leafy spurge (Steenhagen and Zindahl 1979).

Pests—Although many pests of leafy spurge have been identified, none has been shown to effect much control on this weedy species.

Reproduction

Reproduction/sexual—Flowers of leafy spurge are insect pollinated. The flowers produce copious amounts of pollen and nectar. A survey in Saskatchewan showed 8 orders, 39 families, and 60 species of insects on the flowers of leafy spurge (Best et al. 1980).

Fruits ripen and seeds are dispersed from mid- to late-July in the United States. The number of seeds produced per stalk varies from 252 seeds in habitats where spurge competes with native grasses to about 200 seeds where spurge competes with annual weeds and crested wheatgrass (Selleck et al. 1962). Seed yield can be very high. In Saskatchewan, leafy spurge patches were calculated to produce 24 to 3,400 pounds of seed per acre (Selleck et al. 1962).

Seeds of leafy spurge have a rather high germination rate of 60 to 80 percent (Bakke 1936, Bowes and Thomas 1978, Hanson and Rudd 1933). Seed may remain dormant for about 5 to 8 years following maturity, but 99 percent of the germination occurs within the first 2 years (Selleck 1959). The optimal temperature for germination is 30 to 32 degrees Celsius. Alternate freezing and thawing, wet and dry periods, and prolonged dark periods promote germination; scarification does not (Selleck 1959). The peak period for germination is late May to early June, but given adequate moisture, seeds will germinate throughout the growing season.

Seed dispersal is initially affected by explosive dehiscence of the seed capsule. The seed may be ejected up to 4.6 meters from the parent and distributed fairly uniformly from 0.3 to 4.0 meters from the plant (Hanson and Rudd 1933). The seeds can also float and initial infestations often occur along stream or river banks where seeds have floated into appropriate habitat. Birds have been implicated in spreading seed, but documentation is limited except for sharptail grouse.

Reproduction/asexual—One of the most important aspects of leafy spurge biology (in addition to production of large amounts of seed) is its ability to reproduce and spread rapidly via vegetative reproduction. Vegetative reproduction occurs from both crown buds and root buds that overwinter and produce new shoots in the spring. The crown of leafy spurge develops just under the surface of the soil and produces a large number of buds that annually produce new stems. The crown region of the plant can also produce new roots that contribute to the spread and persistence of the plant. Leafy spurge crowns can live for many years, but the number of years is unknown (Bowes and Thomas 1978).

Seedlings have a remarkable capacity for vegetative reproduction and can develop buds within 7 to 10 days after emergence. Buds form on the proximal portion of the seedling's hypocotyl. The number of buds produced on the hypocotyl is limited, unlike the roots where up to six times as many buds will form. Bud formation limits the growth of the seedling. All hypocotylar buds and root buds have the potential to produce a new shoot axis.

Once control practices have been initiated, it is the root system that ensures that leafy spurge spreads and persists in the soil. The root system, consisting of long roots and short roots, can give rise to shoot buds almost anywhere along its length. The long shoots give rise to most of the buds and have been excavated to a depth of 4.8 meters (Best et al. 1980). The upper part of the plant can be killed by herbicides or tillage, but living roots below the treatment zone or detached roots will regenerate new shoots. Cultivation or other shallow removal of leafy spurge plants can actually cause a net increase in the number of stems in an infestation. This was demonstrated by Selleck et al., (1962) who showed that regrowth of leafy spurge after rototilling averaged 316 shoots per square meter in comparison to 134 shoots per square meter in undisturbed control. Shoots can emerge from 90 centimeters of overlying soil for 5 successive years after removal of the major portion of the root system by excavation (Coupland et al. 1955).

Impacts

Euphorbia esula presents a management problem because it is a long-lived, aggressive perennial weed that tends to displace all other vegetation in pasture, rangeland, and native habitats. It is invasive because of the large number of seeds it produces and because it has the capability of producing large numbers of underground shoot buds that can each produce a new shoot. It is particularly aggressive in drier sites, such as hillsides and prairies. Yield reductions of desirable forage species associated with stands of leafy spurge have been reported to decrease from 10 to 100 percent (Reilly and Kaufman 1979). Forbs and grasses in natural areas may be completely displaced by leafy spurge in a few years if the infestation is left unchecked.

Euphorbia esula is rapidly spreading into many areas of Midwestern United States. Control is difficult and must begin before successful establishment or control may become impossible. Rapid reestablishment of dense stands occurs after an apparently successful management effort because of the long-lived root system present in the soil.

Approximately 2.5 million acres are infested with leafy spurge in the U.S. and Canada with the number of infested acres increasing yearly (Dunn 1979). The weed can spread rapidly as evidenced by the doubling of the acreage infested by leafy spurge in North Dakota from 1973 to 1982, a period of 9 years. The Minnesota Department of Transportation estimates that 800,000 acres of land in 80 counties in Minnesota have leafy spurge on it, with the most severe infestations occurring in counties bordering North Dakota.

Management/monitoring

Management requirements

Euphorbia esula is an extremely aggressive and persistent weed that is rapidly spreading into many areas of Midwestern United States. It is invasive because of the large number of seeds it produces and because it has the capability of producing large numbers of underground shoot buds that can each produce a new shoot. If left unchecked, natural areas may become completely overrun with leafy spurge in the period of a few years. It is particularly aggressive in drier sites, such as hillsides and prairies. Control is difficult and must begin before successful establishment, or control may become impossible. Rapid reestablishment of dense stands occurs after an apparently successful management effort because of the long-lived root system present in the soil.

Chemical control

Chemical control, except for continuous tillage or grazing in agricultural situations, is the best method for elimination of leafy spurge. Many herbicides have been used to control leafy spurge with varying degrees of success (Lym and Messersmith 1983). These include picloram, 2,4-D, dicamba, and glyphosate. At the leafy spurge symposium at Montana State University in 1985, it was shown that picloram was the most effective in controlling spurge. Because picloram is expensive, the less costly herbicide 2,4-D is sometimes used alone or mixed with picloram in large areas of spurge infestation. Biannual application of 2,4-D alone would most likely only prevent seed production and spread of spurge with little change in area of original infestation (Lym and Messersmith 1987). Messersmith reported that low rates of picloram with 2,4-D in repeated treatments gave the best long-term control of spurge. Although 2,4-D used alone does not offer as effective control as picloram, it may be preferred in some cases because of its lower cost and perceived lower health risk. One study showed that 2,4-D when used as a set-up treatment for picloram has virtually no effect on control of spurge (Gamal 1986). Dicamba and 2,4-D are often used as followup treatments to picloram, but with mixed and often disappointing results (Bybee 1981, Mitich 1972).

Dicamba has met with some success in the control of spurge, but is costly and breaks down quickly in the soil. Picloram is clearly more effective than dicamba in the eastern portion of spurge country (North and South Dakota, Nebraska, Minnesota, and Wisconsin) because of its longer soil residual activity. However, in Western States, such as Wyoming, Montana, and Colorado, where rainfall is relatively low, dicamba is not leached or broken down as quickly and has been found to give quite effective control. In these States, dicamba may be preferred because of its short soil residual time and corresponding decreased threat to returning desired forbs. Dicamba at high rates (6–8 lb/ac) may decrease production of native grasses. The bluegrasses (Poa spp.) are the most tolerant grasses to dicamba at high rates (Lacey et al. 1985). Picloram at high rates also damages grasses, but it is usually prescribed at relatively low rates for spurge control.

Generally, leafy spurge control with herbicides increases native grass production, although picloram can damage smooth brome and glyphosate applied in the fall severely decreases forage yield (Gylling and Arnold 1985). Picloram and dicamba are restricted from use among trees, and 2,4-D amine or glyphosate is recommended. Care must be taken to avoid contacting tree foliage with herbicide directly or from spray drift.

Two other chemicals that show promise in spurge control and which may be available in the future are flouroxypyr and sulfomenturon.

Leafy spurge is sensitive to the timing of herbicide application, with control being most effective with 2,4-D, picloram or dicamba in mid to late June (seed development) and in late September (fall regrowth) (Lym and Messersmith 1983). Distinction between appearance of bracts and true flowering is important for timing herbicide application. Spring herbicide application is more effective on plants with developing true flowers than on plants with developed bracts, but undeveloped flowers (Eberlein et al. 1982). Glyphosate is most effective when applied after seed set in midsummer or late in September after fall regrowth has started, but before a killing frost (Lacey et al. 1985). Chemical control must be thorough and persist for several, often many, years. Some herbicides must be applied annually or semi-annually. One application of picloram sometimes provides adequate control for several years, but followup applications are often necessary, once in spring to prevent seed development and again in fall to promote translocation of the herbicide to the roots. If infestations are limited and caught early, eradication may be possible. If infestations are severe, it may be difficult to stop the spread of spurge except at great economic and biological expense.

The most widely used or recommended herbicides and their application rates follow:

Picloram—Scattered patches or nearly inaccessible areas of spurge: 2 pounds per acre late spring picloram followed by 2 pounds per acre early fall; the result is 85 to 90 percent shoot control for 3 to 4 years; when shoot control drops below 75 percent, retreat with 0.5 pound per acre. As long as the area is under continuous surveillance, use 1 pound per acre initially for less damage to grasses.

Large, uniform infestations that are accessible and easily treatable on a yearly basis: late spring, 0.5 pound per acre picloram; 70 percent control; must be followed by 0.5 pound per acre once a year.

Picloram is marketed as TORDON®, and in the past has been available in pellet form (Tordon® 2K and Tordon® 10K) and in liquid form (Tordon® 22K, Tordon® K, and Tordon® 101). The pellet forms are no longer available. Tordon® 22K is labeled for range and pasture and consists of picloram at 2 pounds per gallon. Tordon® K is labeled for utility rights of way and forestry and wildlife habitat, but is essentially the same product as Tordon® 22K. Tordon® 101 is a mixture of picloram and 2,4-D at 0.54 + 2 pounds per gallon (Brooks 1987).

Picloram + 2,4-D—A less expensive, but effective treatment is 0.25 pound per acre picloram mixed with 1 pound per acre 2,4-D, applied once a year in the spring. This provides 40 to 60 percent control the first year, and if reapplied on an annual basis, will add about 10 percent control each year until more than 90 percent control is achieved after 4 to 5 years (Lym and Messersmith 1987).

2,4-D—Another inexpensive treatment, but less effective than picloram: 2,4-D low volatile ester, oil- or water-soluble amine formulations applied annually at 1.5 pounds per acre twice a year in mid-June and early to mid-September, or 3 pounds per acre applied once per year in spring or fall.

Among trees use 2,4-D oil- or water-soluble amine at 1.0 to 1.5 pounds per acre applied annually in spring or fall.

Glyphosate—Another treatment that may be used among trees is glyphosate at 0.75 pound per acre, applied from mid-August to mid-September; 80 to 90 percent control, may require followup the next spring with 2,4-D at 0.5 to 1.0 pound per acre (Lacey et al. 1985).

Dicamba—Dicamba at 4 to 8 pounds per acre applied in mid- to late-June provides 50 to 80 percent control the first year, but control usually decreases the second year due to the low residual effectiveness of the herbicide. As mentioned above, dicamba may be more effective in low rainfall areas of the Western States.

Dicamba + 2,4-D at .5 + 1 pound per acre may provide better control than either chemical alone (Gylling and Arnold 1985).

Biological control

There are high hopes for the use of biological control agents in the control of leafy spurge, although none of the insects tested has become well established in the United States. Research is ongoing at several locations on at least 15 insects as possible biocontrol agents for spurge (see Research programs). The most well known and widely studied of these to date has been the spurge hawkmoth (*Hyles euphorbiae*). The moth is native to Southern and Central Europe, northern India and Central Asia and was first introduced in North America in Canada in 1963 (Holloway 1964). Several days after the adult female deposits eggs on leafy spurge plants, small larvae emerge and begin to consume spurge foliage as they proceed through five instars over 2 to 3 weeks. After the fifth instar the larva burrows into the soil and pupates (Forwood and McCarty 1980). *H. euphorbiae* has been released at a number of sites in Montana, North Dakota, and some neighboring States on an almost annual basis since 1964, but the moth does not overwinter well and has not become established at many sites. Lacey et al. (1985) reported good establishment of the hawkmoth at two locations in Montana. Once colonies build to a certain population density they become susceptible to a virus that causes severe mortality, so it is difficult to maintain moth populations at densities sufficient for control of the spurge. Since spurge is also resistant to defoliation, the hawkmoth by itself is not a promising biocontrol agent. One suggestion is that adequate control of leafy spurge requires a combination of several insect control agents that attack different parts of the plant, most likely in conjunction with the use of herbicides or other control methods. The hawkmoth may be valuable as one of these agents (Forwood and McCarty 1980).

Research on other agents is still in screening or early stages of release programs, and results will not be clear for several years. Some of the more promising agents for control of spurge are stem and root borers, such as the cerambycid, *Oberea erythrocephala*, and the clear-winged moth, *Chamaesphecia tenthrediniformis;* the gall midge, *Bayeria capitigena*, which prevents flowering of spurge; and the rust fungus, *Uromyces scutellatus*, which devastates shoots by causing systemic infections (Lacey et al. 1984, Schroeder 1980, Pemberton 1986, Bruckart 1986).

Other control methods

Fire would not be likely to provide adequate control of spurge if used alone because its effect would be on top growth and seeds, and established plants would quickly resprout. However, some reports indicate that fire used in conjunction with herbicides gives better control than herbicide application alone Burning in early May followed by herbicide application in June (just before seed set) might offer adequate control.

Burn and herbicide application tests on leafy spurge were done in South Dakota in 1984 and 1985 with good results. Plots were sprayed with a mix of 2,4-D and picloram in September 1984 and burned the following April, sprayed again in June and burned again in October of 1985. Leafy spurge generally burns well because of its high oil content, but he felt that a herbicide application before burning allowed for an even better fire. Burning reduced seed viability to about 10 percent, and seedling development was greatly reduced in the burned and sprayed plots compared to plots that received either burning or herbicide treatment alone. Plots were located on a flood plain dominated by leafy spurge and silver sage brush (previously existing grasses having been replaced by the spurge) and in upland areas dominated by spurge with grasses, such as needle and thread (*Stipa comata*) and western wheatgrass (*Agropyron smithii*). Two years after treatment, the burned and sprayed plots were still "islands" virtually free from spurge within a larger spurge-infested area, and many of the native grasses and forbs had become reestablished.

Spurge generally presents a problem for chemical control because of its indeterminate growth. Herbicides are developed to be most effective during stages of greatest vegetative growth and in a single stand of spurge at one time some plants may be releasing seed, some in full bloom, and some not yet flowing. The burning treatment alone greatly stimulated vegetative production, and those plots produced a thick, uniform stand of spurge, which appeared ideal for application of herbicides. Thus, burning first followed by herbicide application may also be effectie for spurge control, although this was not tested.

Work using prescribed burning to remove litter and seeds followed by herbicide application has been done in the Lostwood National Wildlife Refuge in North Dakota. The burning either burns up the seeds or scarifies them causing germination. With the litter removed, the newly sprouted seedlings are easily detected and then sprayed with herbicide. The timing of the burn does not seem critical, except that it should be 3 to 4 weeks before herbicide application. At Lostwood, burns are conducted primarily for control of woody species and may be set in mid-June or late summer. Burning must be conducted repeatedly (for example, every other year for 5 to 6 years) to ensure that all seeds are burned or germinated. Herbicide should be applied twice a year, in the spring and fall. Excellent control of spurge resulted with this method.

Repeated mowing or hand cutting may also be used to control seed production, but must be used in conjunction with herbicides for adequate control of stand expansion. Repeated mowing or cutting during a single season is necessary because a single cutting (removal of apical meristem) stimulates the development of inflorescence on lateral branches (Selleck et al. 1962). Mowing also affects grasses and forbs in the mowed area. Since leafy spurge resprouts rapidly, mowing would probably reduce the competitive ability of other species. Selective clipping of the spurge may be preferable, but is time-consuming. For small patches, the use of a hand sickle allows relatively rapid cutting of spurge with little adverse effect on other plants. An automatic "weed eater" works more quickly, but allows for less selectivity than a hand sickle. Clipping the tops to within 4 inches of the ground just before seed set prevents the plants from going to seed. If no herbicide is applied, clipping may be necessary again in midsummer to prevent futher seed development. Without a fall herbicide application, this method may inhibit stand expansion, but is unlikely to reduce spurge abundance in a patch.

Grazing of sheep has been used successfully to control spurge on ranches in Montana, but ranchers agree that once the sheep were removed, the spurge would quickly return (Lacey et al. 1984).

Competition from other plant species may be a means of control in natural areas, but few, if any, plants have been found that show early spring growth, have dense foliage, and are resistant to broadleaf herbicides. The University of Wyoming is conducting interseeding tests with spurge and 10 grass species in tilled and untilled areas in conjunction with herbicide treatment. Preliminary results show that some grasses performed better in tilled and some in untilled areas, but results must be monitored for several years before any conclusions are reached. In Colorado, leafy spurge appears to be limited to low altitude, mesic, mainly riparian habitats. Managers are looking at the effects of allowing the shrubs in riparian zones to encroach and compete with the spurge instead of eradicating spurge and shrubs alike with herbicides.

Nature Conservancy Preserves

North Dakota—Cross Ranch currently has about two dozen small, localized patches of leafy spurge totaling about 3 acres. Management consists of spraying picloram each year in June in open areas (old fields, prairies, railroad edges) at 2 ounces per gallon, and glyphosate in forested areas each year in August or September at 4 ounces per gallon. Infestations are not severe, but each year new patches are discovered. The management goal is to eliminate them before they spread. Contact: Cross Ranch, Hensler, ND 58547, 710-794-9841.

South Dakota—Ordway Prairie began to see leafy spurge in 1980, and for several years it was simply hand chopped. It began to spread, so picloram was applied in pellet form in the spring of 1985 and again in liquid form in the spring of 1986. In 1987, there were just scattered patches, which management is continuing to chop by hand. Abundance of flowering stems is down by 80 percent since 1985. If necessary, 2,4-D is applied as a followup treatment. Contact: Ordway Prairie, Star Route 1, Box 16, Leola, SD 57456, 605-439-3475.

Altamont Prairie was acquired in 1963 and no active management was employed for the next 7 years. Leafy spurge was already becoming a severe problem in 1970, the first year of active spurge control. 2,4-D was sprayed in the spring of 1970 and again in 1972 and then every year since 1977. In 1974 and 1976, spurge hawkmoths (*Hyles euphorbiae*) were released, but did not become established. Beginning in 1982, picloram has been sprayed every year in the areas of heaviest spurge infestations. Burns were conducted in late May of 1984, 1985, and 1986. Spurge continues to be a severe problem at Altamont.

Crystal Springs is a new Nature Conservancy acquisition in South Dakota at which there are patches of leafy spurge. Monitoring and mapping of patches were conducted in 1987, and control methods, probably application of picloram, will subsequently be implemented.

Nebraska—At Niobrara Valley Preserve, 5 to 6 acres of grassland and several hundred acres of woodlands have areas of spurge infestations. In 1986, all the nonforest sites were treated with picloram 2K pellets, and picloram liquid was reapplied in 2 to 3 years when shoot control began to decline. No control measures have been implemented in the wooded lands, and the spurge in these areas is difficult to combat. Contact: Niobrara Valley Preserve, Rt. 1, Box 358, Johnstown, NE 69214, 402-722-4440.

Iowa—The largest area of spurge infestation on Nature Conservancy land is on the Sioux City Preserve. Management applied picloram liquid by hand (herbicide dripped directly onto individual stems) to open area infestations in the spring of 1986 and 1987. An experimental mowing treatment is planned for one large patch in a disturbed area. The entire area will be mowed every few weeks over two seasons, and herbicide (picloram) will be applied the third season. Management is also planning a burning treatment in conjunction with the herbicide in the other grassland areas. Hillsides, which have small patches of spurge, will be burned in the fall and followed with picloram spot application in the spring. Lowland areas, where spurge infestations are the greatest, will be burned in the spring and followed with picloram application. The largest infestations, as at Niobrara Valley, are in wooded areas where no control measures have been implemented. Contact: TNC Iowa Field Office, 424 10th St., Suite 311, Des Moines, IA 50309, 515-244-5044.

Minnesota—Minnesota's biggest spurge problem is at Bluestem Prairie in the northwestern corner of the State. In 1984, four patches were known. Intensive searching revealed 50 patches by the end of 1985, 104 patches in 1986, and over 177 patches by 1987. Many small patches have grown together since 1985. Data collected on 10 individual patches showed about 129 percent increase in patch size over 2 years. Management applied picloram to the most severe infestations, and many areas were burned or mowed in addition to the herbicide application. Spurge is also a management concern on several other Minnesota preserves. Contact: TNC MN Field Office, 1313 5th St. SE, Minneapolis, MN 55414, 612-379-2134, Non-Nature Conservancy Lands.

This is not a complete list of areas currently under management for control of leafy spurge. The information and contacts are included because they are examples of successful and/or informative treatment programs.

Devil's Tower National Monument in Wyoming has been applying herbicides to spurge-infested areas for about 20 years. Years ago there were large infested patches over the entire 14,000-acre park, and spraying was conducted by truck on a relatively large scale. 2,4-D was used initially until picloram was discovered to be more effective. There are no longer large patches of spurge so picloram (22K liquid at 1 gal/ac) is spot sprayed on individual plants by an employee who walks the ground with a backpack sprayer throughout the season. These procedures have successfully reduced the abundance of spurge to scattered patches, but herbicide treatment will continue indefinitely to prevent spread and reinfestation. Contact: Maintenance Foreman, Devil's Tower National Monument, Devil's Tower, WY 82714, 307–467–5603.

The Custer National Forest has more than 8,000 acres of spurge infestations, the major areas are in the Sheyenne District in North Dakota. Management has been applying picloram annually at 0.25 to 1 pound per acre. The number of infested acres has not so far decreased, but spread is being prevented except in drainage ditches that are restricted from use of picloram. Contact: Resource Assistant, Sheyenne National Grassland, Box 946, Lisbon, ND 58054, 701-683-4342.

The Lostwood National Wildlife Refuge in North Dakota has significantly reduced the abundance of leafy spurge with burning plus herbicide application. Burning is prescribed mainly for control of woody species and is conducted in mid-June or late summer every other year (there is not enough fuel to burn every year). Picloram has been sprayed twice a year at recommended rates in mid-June and September since 1979. Herbicide application is always conducted within 3 to 4 weeks following a burn, after the surviving seeds have germinated and sprouted shoots. Areas on the refuge that were once spurge infested no longer have spurge. Contact: Lostwood Refuge Manager, Lostwood National Wildlife Refuge, 701-848-2722.

Roosevelt National Park has at least 700 acres of leafy spurge infestations. Management has been spot spraying picloram since about 1975, and this has been effective at maintaining or decreasing abundance on localized areas, but park-wide, the spurge population has been steadily increasing. The park was an experimental site for release of two biocontrol agents for spurge: the flea beetle *Apthona flava* and the gall midge, *Bayeria capitigen*. These agents were released in experimental plots in 1987. Contact: Theodore Roosevelt National Park, P.O. Box 7, Medera, ND 58645, 701-623-4466.

Monitoring requirements

Monitoring of *Euphorbia esula* on preserve lands is essential to track the success of control practices. Since the root system is extensive and persistent, new shoots can emerge even after complete eradication of aboveground tissues. Monitoring and repeat control measures are generally considered necessary for at least 10 years following initiation of active management.

Monitoring can be accomplished through aerial photography (see below) and ground observation (e.g., during herbicide application). Spurge is most easily detected when in flower from late May to late June. Patches should be carefully surveyed and mapped annually.

Monitoring programs

The U.S. Forest Service is currently researching protocol for aerial photography of *Euphorbia esula*. Studies are being conducted to determine the best scale (1:16,000, 1:24,000) film type and season in which to conduct aerial surveys, and a user's handbook is in production. Contact: U.S. Forest Service, Forest Pest Management-Methods Application, Fort Collins, CO, 303-224-1785.

The University of Nebraska is marketing a highly sophisticated image processing and analysis package, which can be used in conjunction with aerial photography to map spurge infestations and other important species. Contact: MicroImages Inc., 932 Lakeshore Drive, Lincoln, NE 68529, 402-435-3864.

Research

Management research programs

Researchers at North Dakota State University are investigating about 15 insects as potential biocontrol agents for *Euphorbia esula*. Particularly promising agents include several stem and root borers, a gall midge that stops flowering, and a larva that consumes seeds prior to dispersal. Various herbicides and application rates are also being continuously tested. A new chemical being tested at NDSU that looks promising for control of spurge is sulfomenthuron. The Cooperative Extension Service at NDSU publishes a leafy spurge newsletter four to five times a year. Contact: Editor, Leafy Spurge News, 1924 North Grandview Lane, Bismarck, ND 58501, 701-663-6445; or Department of Agronomy, North Dakota State University, Fargo, ND 58105, 701-237-7971.

Research on biocontrol and herbicide use is also being conducted at South Dakota State University. Contact: Extension weed specialist, Plant Science Department, South Dakota State University, Brookings, SD 57007, 605-688-5121.

At the University of Wyoming, research is being carried out on chemical control, interseeding of spurge with grasses, mowing, and biocontrol. A new chemical that has been found effective in controlling spurge and that shows excellent grass tolerance is flouroxypyr. Interseeding studies are showing that some grasses may be good competitors against spurge when used in conjunction with herbicides (such as flouroxypyr). Contact: Extension specialist–weed science, P.O. Box 3354, University Station, Laramie, WY 82071, 307-766-3115; or Department of Entomology, P.O. Box 3354, University Station, Laramie, WY 82071, 307-766-5199.

Research at the University of Minnesota is focused on spurge development and allelopathic inhibition of seed germination. Contact: Department of Botany, 220 BioScience Center, University of Minnesota, St. Paul, MN 55108, 612-376-1558.

In Montana, research has been ongoing for several years. Current projects include grazing of spurge with sheep and goats and biological control. Contact: Department of Plant and Soil Science, Montana State University, Bozeman, MT 59717, 406-994-5061; or Department of Entomology, Montana State University, Bozeman, MT 59717, 406-994-6405.

Management research needs

Integrated pest management should be a priority research area for *Euphorbia esula*. Control of spurge is a complex problem that requires an integrated approach. The use of herbicides has helped reduce spurge abundance and/or prevent its spread in many areas, but the chemicals must be reapplied, often on an annual basis, for an indefinite number of years to maintain control and prevent recurrences. Nonchemical controls would reduce the dependence on biocides and might help in areas where use of picloram, the most effective chemical, is restricted (e.g., in forests and wetlands).

Control of *Euphorbia esula* in wooded areas is a particular problem at several Nature Conservancy preserves, notably Niobrara Valley in Nebraska and Sioux City in Iowa, and effective management programs are lacking in these areas. The goal of research should be to develop effective control programs for leafy spurge that make use of a variety of methods (i.e., biological control, mowing, burning) and would allow for a gradual reduction in herbicide applications.

More information is also needed on *Euphorbia esula* biology, particularly in the areas of plant development and ecology. Questions of interest include the following:

- What are the germination requirements of leafy spurge, and how are seeds dispersed?
- How does crown bud and root bud development proceed?
- Would application of any plant growth substance inhibit vegetative reproduction?
- Can cold hardening of crown buds in the fall be prevented such that the crown would not survive over winter?
- Can any desirable grasses or broadleaf species be managed to outcompete and displace spurge in certain areas?

Bibliography

Bakke, A.L. 1936. Leafy spurge, *Euphorbia esula* L. Iowa State Agricul. Exp. Sta. Res. Bul. 198:207–246.

Batho, B. 1931. Leafy spurge. Manitoba Dep. Agricul. and Immigration Cir. 106, 4 pp.

Best, K.F., G.G. Bowes, A.G. Thomas, and M.G. Maw. 1980. The biology of Canadian weeds:39, *Euphorbia esula* L. Canadian J. Plant Sci. 60:651–663.

Bowes, G.G., and A.G. Thomas. 1978. Longevity of leafy spurge seeds in soil following various control programs. J. Range Mgt. 31:137–140.

Bruckart, W.R. 1986. Progress and prospects for plant pathogens to control leafy spurge. Leafy Spurge News, R. Lorenze, ed., Agric. Exp. Sta. Coop. Ext. Serv. NDSU. Fargo, ND 7(2): 5–6.

Bybee, T.A. 1981. Leafy spurge control and reestablishment after herbicide treatment. Ph. D. thesis, North Dakota State Univ., 88 pp.

Coupland, R.T., and J.F. Alex. 1955. Distribution of vegetative buds on underground parts of leafy spurge (*Euphorbia esula* L.). Canadian J. Agricul. Sci. 35:386–405.

Dunn, P.H. 1979. The distribution of leafy spurge (*Euphorbia esula*) and other weedy *Euphorbia* spp. in the United States. Weed Sci. 27:509–516.

Eberlein, C.V., R.G. Lym, and C.G. Messersmith. 1982. Leafy spurge identification and control. North Dakota Ag. Exp. Sta. Circ. w-765, 4 pp.

Forwood, J.R., and M.K. McCarty. 1980. Control of leafy spurge (*Euphorbia esula*) in Nebraska with the spurge hawkmoth (*Hyles euphorbiae*). Weed Sci. 28(3):235–240.

Gamal, H. 1986. The use of 2,4-D as a set-up treatment prior to light rates of picloram for leafy spurge shoot control. Proc. Leafy Spurge Conf., Bozeman, MT.

Gylling, S.R., and W.E. Arnold. 1985. Efficacy and economics of leafy spurge (*Euphorbia esula*) control in pastures. Weed Sci. 33:381–385.

Hanson, H.C., and V.E. Rudd. 1933. Leafy spurge life history and habits. North Dakota Agricul. Col. Exp. Sta. Bul. 266, 24 pp.

Holloway, J.K. 1964. Projects in biological control of weeds. *In* DeBach, P., ed. Biological Control of Insect Pests and Weeds, Chapman and Hall Ltd., London, pp. 656–600.

Lacey, C.A., P.K. Fay, R.G. Lym, C.G. Messersmith, B. Maxwell, and H.P. Alley. 1985. Leafy spurge distribution, biology, and control. Montana State U. Coop. Ext. Serv. Circ. 309.

Lacey, C.A., R.W. Kolt, and P.K. Fay. 1984. Ranchers control leafy spurge. Rangelands 6(5):202–204.

Lym, R.G., and C.G. Messersmith. 1983. Control of leafy spurge with herbicides. North Dakota Farm Res. Bul. 40 (5):16–19.

Lym, R.G., and C.G. Messersmith. 1987. Leafy spurge control and herbicide residue from annual picloram and 2,4-d application. J. Range Mgmt. 40 (3):194–198.

Mitich, L.W. 1972. Three years leafy spurge control trials with herbicides. Res. Rep., North Central Weed Contr. Conf. 29:32–33.

Pemberton, R. 1986. Biological control update. Leafy Spurge News, R. Lorenz, ed., Ag. Exp. Sta. Coop. Ext. Ser. 7 (1):3.

Reilly, W., and K.R. Kaufman. 1979. The social and economic impacts of leafy spurge in Montana. *In* Proc. Leafy Spurge Sump. North Dakota Coop. Ext. Serv. Fargo, ND, pp. 21–24.

Selleck, G.W. 1959. The ant ecology of *Euphorbia esula* L. Ph.D. thesis, Dep. Botany, Univ. Wisconsin, 309 pp.

Selleck, G.W., R.T. Coupland, and C. Frankton. 1962. Leafy spurge in Saskatchewan. Ecological Monog. 32:1–29.

Shroeder, D. 1980. Investigations on Oberea erythrocephala (Shrank) (Col;Cerambycidae), a possible biocontrol agent of leafy spurge Euphorbia spp. (Euphorbiaceae) in Canada. Zeit. fur ang. Ent. 90:237–254.

Steenhagen, D.P., and R.L. Zindahl. 1979. Allelopathy of leafy spurge (*Euphorbia esula*). Weed Sci. 27:1–3.

To Contents

III.J.2.c Control and management of purple loosestrife (*Lythrum salicaria* L.) (LYSA2)

(J. Bender, The Nature Conservancy, Arlington, Virginia, author; Jay Rendall, The Nature Conservancy, Arlington, Virginia, update January 11, 1987, and revised 1998; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001. Distribution Map and photographs, NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

This abstract provides information on the biology and control of invasive plant species in wetlands.

To the user

Element Stewardship Abstracts (ESAs) are prepared to provide The Nature Conservancy's Stewardship staff and other land managers with management-related information on those species and communities that are most important to protect, or most important to control. The abstracts organize and summarize data from numerous sources including literature and researchers and managers actively working with the species or community.

We hope, by providing this abstract free of charge, to encourage users to contribute their information to the abstract. This sharing of information will benefit all land managers by ensuring the availability of an abstract that contains up-to-date information on management techniques and knowledgeable contacts. Contributors of information will be acknowledged within the abstract and receive updated editions.

For ease of update and retrievability, the abstracts are stored on computer at the national office of The Nature Conservancy. This abstract is a compilation of available information and is not an endorsement of particular practices or products.

Please do not remove this cover statement from the attached abstract.

Contents

The Nature Conservancy
Element Stewardship Abstract
Purple Loosestrife (*Lythrum salicaria*)

Identifiers

Common name: Purple Loosestrife

General description

Lythrum salicaria is a stout, erect perennial herb with a strongly developed taproot. The plant ranges in height from 0.5 to 2.0 meters. The four-angled stem can be glabrous to pubescent. The sessile leaves are opposite or in whorls, lanceolate to narrowly oblong, with cordate bases. The inflorescence is spike-like, 1 to 4 decimeters long. Petals, five to seven, usually magenta, but white or light pink flowers are also common. The flowers are trimorphic concerning the relative lengths of the stamens and style. The fruit is a capsule, with small seeds, each weighing 0.06 milligrams (Balogh 1985, Rawinski 1982, Gleason 1952, Fernald 1950).

At a distance, *L. salicaria* may be confused with *Epilobium angustifolium*, *Verbena hastata*, *Teucrium canadense*, or *Liatris* spp. Upon closer examination, however, purple loosestrife is easily distinguished from these other magenta-flowered plants.

Stewardship summary

Monitor natural areas for the presence of *L. salicaria*. Maintain preserves so that purple loosestrife cannot invade and flourish. For small infestations, eradication is possible with spot applications of glyphosate herbicides. Monitor the containment and control procedures.

Current methods for eradicating large, dense populations of loosestrife are not totally effective. Mechanical control methods are ineffective, and the herbicide most effective is nonselective. Realistically, the long-term control of large populations may require biological controls and/or better herbicides, but their development is at least several years away. Therefore, containment and minimizing seed production are the present control objectives for large, dense populations. (MN DNR 1987)

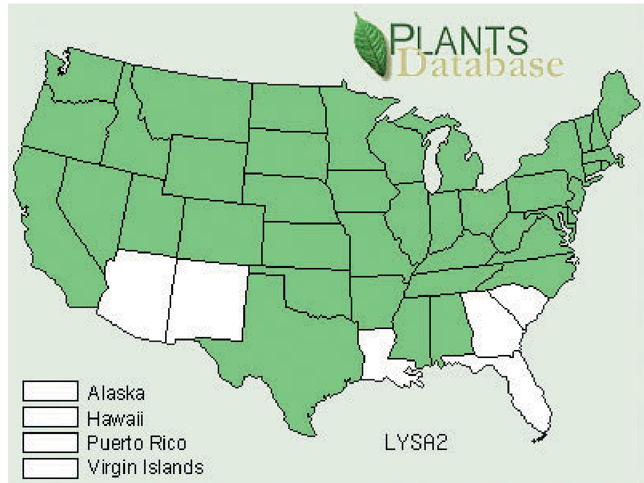
Natural history

Habitat

L. salicaria is native to Eurasia and was first reported from the northeastern coast of North America in 1814 (Stuckey 1980). Although purple loosestrife occurs in nearly all sections of the United States, the heaviest concentrations are in the glaciated wetlands of the Northeast. Occurrences west of the Mississippi River appear to be scattered (Stuckey 1980), with the species establishing in reclamation projects in the West (Thompson and Jackson 1982).

Purple loosestrife is found in wetlands, such as cattail marshes, sedge meadows, and open bogs. *L. salicaria* also occurs along streambanks, riverbanks, and lakeshores. In addition, the plant is found in ditches and other disturbed wet soil areas.

L. salicaria grows best in high organic soils, but tolerates a wide range of soils including clay, sand, muck, and silt (Thompson and Jackson 1982). Generally, the plant is found in full sun, but it can survive in 50% shade (Thompson and Jackson 1982). Typical associates include *Typha latifolia*, *T. glauca*, *Phragmites australis*, *Spartina* sp., *Scirpus* spp., and *Carex* spp. (Thompson and Jackson 1982).



Reproduction

Purple loosestrife begins to bloom in July and continues until September or October. The flowers are pollinated by several types of bees (*Megachilinae*, *Apinae*, *Xylopinae*, and *Bombinae*) and by several butterflies (*Pieris rapae*, *Colias philodice*, and *Cercyonis pegala*) (Balogh 1985). Seed production is prolific. Each capsule averages 120 seeds, and each plant has up to 900 capsules (Rawinski 1982). The lowest capsules on the stem are dehiscing while the upper stem capsules are still green.

The seeds are small, weighing 0.06 milligrams each (Shamsi and Whitehead 1974). Dispersal is mainly by wind, but seeds can also be transported on the feet of waterfowl or other wetland animals. Red-winged blackbirds have been observed eating the seeds (Rawinski 1982). Humans carry seeds inadvertently on clothing and shoes and in some instances, beekeepers have purposely sown seeds in headwaters and wetlands to provide a steady source of nectar for their bees. The seeds and cotyledon stage seedlings are buoyant and can be dispersed by water currents (Balogh 1985). The seedbank potential for *L. salicaria* is enhanced by the high viability of the seeds. Viability decreased from 99 to 80 percent after 2 years of storage in a natural body of water (Rawinski 1982).

Seeds of *L. salicaria* can germinate in acidic or alkaline soils; in soils that are nutrient rich or nutrient poor. Light requirements for germination are minimal (Shamsi and Whitehead 1974). Temperature at the soil surface is a critical factor for germination. Seeds germinate at temperatures ranging from 15 to 20 degrees Celsius (Balogh 1985). Seeds germinate in high densities—about 10,000 to 20,000 per square meter (Rawinski 1982). The interval between germination and flowering is 8 to 10 weeks (Rawinski 1982).

Seedlings that germinate in the spring grow rapidly and produce a floral shoot up to 30 centimeters in length the first year. Summer-germinated seedlings develop only five or six pairs of leaves before the end of the growing season (Shamsi and Whitehead 1974). Spring-germinated seedlings have a higher survival rate than that of summer-germinated seedlings. Open grown shoots have a greater reproductive output than shoots growing in dense stands (Rawinski 1982). Once established, seedlings can survive shallow flooding from 30 to 45 centimeters in depth (Thompson and Stuckey 1980.).

The taproot is strongly developed in the seedling stage and persists throughout the life of the plant (Shamsi and Whitehead 1974). In mature plants, the taproot and major root branches become thick and woody (Rawinski 1982). The semi-woody aerial shoots die in the fall, but persist for 1 to 2 years making stands of *L. salicaria* very dense. New shoots arise the following spring from buds at the top of the rootstocks (Rawinski 1982).

The rootstock is the main organ of perennation, and vegetative spread is therefore limited (Shamsi and Whitehead 1974). *L. salicaria* can spread vegetatively by resprouting from cut stems and regenerating from pieces of rootstock (Rawinski 1982).

Infestations of purple loosestrife appear to follow a pattern of establishment, maintenance at low numbers, and then dramatic population increases when conditions are optimal. *L. salicaria* flourishes in wetland habitats that have been disturbed or degraded from draining, natural drawdown in dry years, bulldozing, siltation, shore manipulation, cattle trampling, or dredging. Mudflats exposed following drawdowns are quickly colonized if a loosestrife seed source is present. Seeds are usually present in such large numbers and germinate in such high densities that growth of native seedlings is suppressed (Rawinski 1982). Loosestrife crowds or shades out native species and eventually becomes a virtually monospecific stand.

L. salicaria is an extremely successful invader of wetlands that have been subjected to some type of disturbance: drawdown, siltation, drainage, ditching. Expansion in a wetland can be extensive and sudden because of the abundance of seeds produced and the rapid growth of seedlings. High seed viability and prolific seed production can build up a seedbank of massive proportions.

Purple loosestrife seed germinates in such high densities that it outcompetes native seedlings. The buildup of debris around the roots enable loosestrife to invade deeper water and to form dense stands that shade out other emergents and push out floating vegetation by closing open water spaces.

Management/monitoring

Management requirements

Once purple loosestrife becomes established in a wetland, it displaces endemic vegetation through rapid growth and heavy seed production (Rawinski 1982). *L. salicaria* has a detrimental impact on native wetland vegetation and associated wildlife. Important wildlife food plants, such as cattails and pondweed, are displaced or shaded out as *L. salicaria* expands across a wetland. If purple loosestrife is left unchecked, the wetland eventually becomes a monoculture of loosestrife (Rawinski 1982). The invasion of *L. salicaria* leads to a loss of plant diversity, which also leads to a loss of wildlife diversity.

Management objectives may include eradicating populations, containing populations, or preventing establishment. Monitoring should be used to track the accomplishment of these objectives.

The best time to search for purple loosestrife is in July and August when the plants are blooming. The bright magenta flowers are easy to spot at a great distance. Aerial surveys can be used to note the yearly position of large populations. An advancing or receding boundary would be identifiable from air photos. Ground surveys are more feasible for tracking small populations and finding newly established populations. Look for seedlings in June.

Control methods

Several control methods have been attempted with varying degrees of success. Natural area managers must determine their objectives first. Is it more feasible to contain or control populations of purple loosestrife? Large populations extending over at least 3 acres are difficult if not impossible to completely eradicate using presently known methods. These large populations should be contained at their present position. Preventing the expansion can be accomplished through hand-pulling new plants along the periphery or spraying herbicide on plants extending beyond the main body of the population. Smaller populations can be controlled through eradication. Populations up to 3 acres can be cleared with herbicides or hand-pulled, depending upon the size of the work crew and the time available.

Chemical—The herbicide glyphosate is most commonly used to control *L. salicaria.* Glyphosate is available under the trade names Roundup™ and Rodeo™, manufactured by Monsanto. Roundup™ cannot be used over water. Another formulation of glyphosate known as Rodeo™ contains a non-ionic surfactant and has been approved for use over water. Ortho X-77 is the non-ionic surfactant recommended for use with Rodeo™, but several other non-ionic surfactants were cleared for use with Rodeo™ in 1985 (Balogh 1985).

The major disadvantage in using Rodeo™ is that glyphosate is a nonspecific systemic. Broadcast spraying of nonselective herbicides kills all of the vegetation and may result in an increase in loosestrife density because of seed germination following the removal of competing perennial vegetation (Minnesota DNR 1987). Spot application of Rodeo™ directly onto *L. salicaria* ensures that no large holes will appear in the marsh vegetation and that competition will be unaffected. The safest method of applying glyphosate herbicide is to cut off all stems at about 6 inches and then paint or drip onto the cut surface a 20 to 30 percent solution (Henderson 1987).

Spraying should be done after the period of peak bloom, usually late August (Balogh 1985, Rawinski 1982). One to two percent solutions of Rodeo™ have been recommended as sufficient to kill *L. salicaria* (Henderson 1987, Minnesota DNR 1987, Balogh 1985, Thrune pers. comm.). Work done by Jim Reinartz at the University of Wisconsin Milwaukee Field Station indicates it is best to spray no more than 25 to 50 percent of a plant's foliage (Henderson 1987). This helps protect against overspraying that might damage adjacent vegetation.

Followup of any control effort is critical in the same growing season and for several years afterwards because some plants will be missed, new seedlings may sprout from the extensive seedbank, and a few plants will survive the low-dosage treatment (Henderson 1987, Minnesota DNR 1987). Higher dosage and careless application, however, inevitably kills more surrounding vegetation and leads to establishment of loosestrife seedlings (Minnesota DNR 1987).

For larger infestations where spot application of glyphosate is not practical, broadleaf herbicides can be used. They have the advantage of not harming monocot species, which are the dominants in most wetland types. Broadleaf herbicides (2,4-D based) can be effective on loosestrife if applied in late May or early June (Henderson 1987). The disadvantage of treating early in the season is that purple loosestrife plants are easily overlooked when not in flower. A combination of 2,4-D and dicamba has had limited use in Western States irrigation ditches (Jackson pers. comm.). The EPA approved a 1:1 tank mix of these two products. Once *L. salicaria* has reached 10 to 15 percent of its mature growth, it can be sprayed with good results. To ensure complete coverage and compensate for spotty application, repeat the treatment once during the growing season (Jackson pers. comm.).

Pulling—Hand-removal is recommended for small populations and isolated stems. Ideally, the plants should be pulled before they have set seed. The entire rootstock must be pulled because regeneration from root fragments is possible. Minimize disturbances to the soil and native vegetative cover. Remove uprooted plants and broken stems from the area since the broken stems can resprout (Rawinski 1982).

Replacement—Replacement control has been attempted in several wildlife refuges (Balogh 1985, Rawinski 1982). Rawinski (1982) sowed Japanese millet (*Echinochloa frumentacea*) with *L. salicaria* and found that the millet seedlings outcompeted the loosestrife seedlings. The millet must be planted immediately after marsh drawdown has occurred. Balogh (1985) found that Japanese millet does not regenerate well and must be replanted every year. He attempted a replacement treatment using native seed. *Polygonum lapathifolium* was seeded with purple loosestrife and the *Polygonum* outcompeted the loosestrife. However, the following spring *L. salicaria* started growing first because of its overwintering rootstock. Replacement methods would have a limited application within a natural area, but they may be useful to control or contain loosestrife populations on buffer property.

Biological—Several characteristics of *L. salicaria* make it an ideal candidate for biological control (Thompson et al. 1987). Batra et al. (1986) completed detailed ecological and host-specificity studies for six European species: a cecidomyiid fly whose galling can reduce purple loosestrife foliage by 75 percent and seed production by 80 percent; a stem and root boring weevil; two chrysomelids that can cause nearly 50 percent defoliation; and two weevils that mine ovaries and seeds. The results of these studies indicated that the chances of successful biological control of *L. salicaria* in North America are excellent.

Research

Management research programs

A research project in Wisconsin includes investigations on different methods of control and different herbicide treatments. The ecology of *L. salicaria* including seedbank buildup is also under investigation. Contact Natural Area Management, 2845 Timberlane, Verona, Wisconsin 53593.

Hand cutting purple loosestrife and fertilizing cattails under varying degrees of wetness is being studied at Indiana Dunes National Lakeshore. Contact Division of Science, 1100 N. Mineral Springs Rd., Porter, Indiana 46304.

A research project funded by the Minnesota Metropolitan Council was conducted by Hennepin County Park Reserve. Chemical control techniques are to be evaluated for 2 years in control plots. Contact Hennepin County Park Reserve, 3800 Co. Rd. 24, Maple Plain, Minnesota 55359.

The Minnesota Legislative Commission on Minnesota Resources has funded a comprehensive control program over a 2-year period. The program will inventory purple loosestrife in Minnesota, keep abreast of current control methods and research, implement a prioritized control program, monitor environmental impact and effectiveness of control, promote public awareness campaigns, and coordinate agencies control efforts within the State. Contact Purple Loosestrife Program, Minnesota Department of Natural Resources, Box 25, 500 Lafayette Rd, St. Paul, Minnesota 55155.

Management research needs

Biological control methods should be a priority for research. Repeated chemical treatments are costly, and the long-term effects on natural systems are not fully understood. Preliminary investigations in Europe revealed several host-specific insects that keep *L. salicaria* in check. Further research is warranted. Research is needed to assess the potential productivity of the seedbank. How extensive is the seedbank in a wetland in comparison to the size of the aboveground population? What is the rate of seed buildup? Can the age of a seedbank be determined? What is the viability of purple loosestrife seed? More research is needed on herbicide treatments that will give the most selective application with the least impact to the surrounding competitive vegetation; i.e., wick applications. Available information suggests that research on mechanical treatments will not yield helpful results.

Bibliography

Balogh, Greg. 1985. Ecology, distribution, and control of purple loosestrife in northwest Ohio. Annual report from October 1984 to September 1985, Cooperative Wildlife Research Unit, Ohio State Univ.

Batra, S.W.T., D. Schroeder, P.E. Boldt, and W. Mendl. 1986. Insects associated with purple loosestrife (*Lythrum salicaria* L.) in Europe. Proc. Entomol. Soc. Wash. 88:748–459.

Fernald, M.L. 1950. Gray's manual of botany, 8th ed. American Book Company, New York.

Gleason, H.A. 1952. The new Britton and Brown illustrated flora of the Northeastern U.S. and adjacent Canada. New York Botanical Garden, New York.

Harper, Bonnie. 1986. Purple loosestrife coalition. Eden Prairie, Minnesota. (Telephone conversation with J. Bender, TNC, MRO.)

Henderson, Richard. 1986. Consultant, Natural Areas Management, 2845 Timberlane, Verona, Wisconsin 53593.

Henderson, Richard. 1987. Status and control of purple loosestrife in Wisconsin. Research management findings, Number 4, Bur. Research, Wisconsin DNR, Madison, Wisconsin.

Jackson, Tom. 1986. Leader, Field Research Station, U.S. Fish and Wildlife Service, Denver, Colorado 80225. (Telephone conversation with J. Bender, TNC, MRO.)

Minnesota Department of Natural Resources. 1987. Control and eradication of purple loosestrife. Unpublished paper prepared by the purple loosestrife program, November 1987.

Notestein, Anne. 1986. The spread and management of purple loosestrife (*Lythrum salicaria* L.) in Horicon National Wildlife Refuge, Wisconsin. M.S. thesis, Univ. Wisconsin, Madison.

Rawinski, Tom. 1982. The ecology and management of purple loosestrife (*Lythrum salicaria* L.) in central New York. M.S. thesis, Cornell Univ.

Schwegman, John. 1986. Director, Botany Program. Illinois Department Conservation, Springfield, Illinois 62706. (Telephone conversation with J. Bender, TNC, MRO.)

Shamsi, S.R.A., and F.H. Whitehead. 1974. Comparative eco-physiology of *Epilobium hirsutum* L. and *Lythrum salicaria* L. I. General biology, distribution, and germination. J. Ecol. 62:279–290.

Stuckey, R.L. 1980. Distributional history of *Lythrum salicaria* (purple loosestrife) in North America. Bartonia 47:3–20.

Thompson, D.Q. and R.L. Stuckey. 1980. Spread, impact, and control of purple loosestrife (*Lythrum salicaria*) in North American wetlands. Unpublished report.

Thompson, D.Q., and T.P. Jackson. 1982. Purple loosestrife alert. U.S. Fish and Wildlife Res. Info. Bul. No. 82-24, Fort Collins, Colorado.

Thompson, D.Q., R.L. Stuckey, and E.B. Thompson. 1987. Spread, impact, and control of purple loosestrife (*Lythrum salicaria*) in North American wetlands. U.S. Fish Wildl. Serv., Fish Wildl. Res. 2, 55 pp.

Thrune, Bill. 1986. Assistant manager, Horicon National Wildlife Refuge. (Telephone conversation with J. Bender, TNC, MRO.)

To Contents

III.J.2.d Emerging themes in reed canarygrass management (*Phalaris arundinacea* L.) (PHAR3)

(Clayton J. Antieau (author), wetland biologist/botanist/horticulturist, Washington State Department of Transportation, Seattle, Washington, July 2000; edited by Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland December 2001. Photographs and map by NRCS Plant Data Center, Baton Rouge, Louisiana.)

Purpose

Reed canarygrass (*Phalaris arundinacea* L.) (PHAR3) poses serious challenges to management and restoration of riparian and wetland resources throughout the Northern United States. This aggressive grass displaces desirable habitat and species diversity while persisting in the face of active weed control efforts. A comprehensive, multifaceted approach to managing this species is required. This paper reviews practical considerations in managing reed canarygrass. Available management tools include mowing, herbicide application, grazing, cultivation, prescribed burning, flaming, micronutrient management (boron), macronitrogen management, shading and competitive exclusion, flooding, mechanical barriers, and bombing. Several lessons emerge from our experiences in managing this grass. These include a need to understand the importance of integrated approaches, regular monitoring, followup weed control efforts, and the implementation of new technologies and strategies (adaptive management). An information clearinghouse is needed where interested parties can share their management experiences with this weed or seek information resulting from others' experiences.

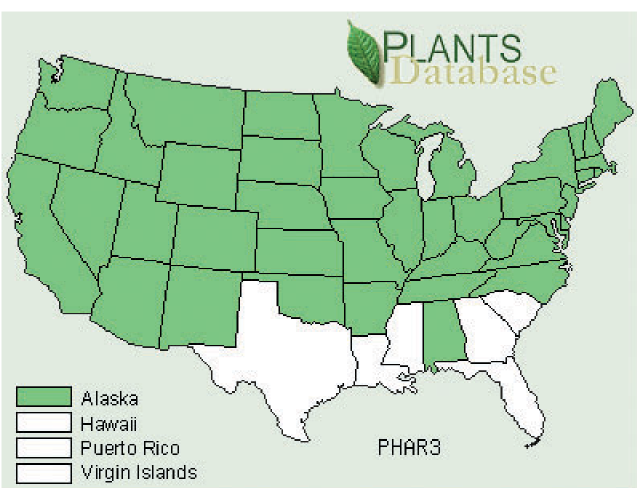
Contents

The restoration of aquatic and terrestrial ecosystems relies strongly on effective management of reed canarygrass (*Phalaris arundinacea*) when this species is present. This paper presents an overview of the conservation and weed management concerns involving reed canarygrass. This overview describes aspects of this species' biology as they pertain to its management as a weed. Naglich (1994) provides a more detailed biological summary and additional description of techniques used historically in managing this species as a weed.

Reed canarygrass is a long-lived perennial, cool-season grass native circumboreally to the Northern Hemisphere (Merigliano and Lesica 1998). An intriguing aspect of this "native status" description involves the possibility that indigenous populations of reed canarygrass have been supplemented by a more aggressive germplasm introduced from Europe (Guard 1995, Galatowitsch et al. 1999). This species is a vigorous, tall-growing plant with an aggressive underground stem (rhizome) system. As a mature plant, reed canarygrass tolerates prolonged soil saturation and ponding as well as dry soil conditions, but is intolerant

of deep shade (Forman et al. 2000). Farmers in the Pacific Northwest consider it to be one of the best forage grasses for wet situations, and it has been and is currently widely planted and promoted for use as a forage/hay crop and in erosion and sedimentation management. The recent development of low-alkaloid cultivated varieties of reed canarygrass has made this species even more attractive for agricultural use in Washington and Oregon.

From an ecological perspective, reed canarygrass competitively displaces desirable native plant species and limits biodiversity in wetland and riparian communities. These changes most likely precipitate effects on other wetland and riparian functions, such as wildlife habitat. Reed canarygrass also evapotranspires large quantities of soil moisture and potentially affects hydrologic characteristics. This species' aggressive growth and high biomass production affects hydraulic characteristics of surface water by clogging ditches and streams with thick thatch and wrack. In many cases infestations appear to form neo-climax communities (*sensu* Daubenmire 1968). These are plant communities that arise through human-caused perturbations and that subsequently prevent original climax vegetation from reestablishing, except in the face of new perturbation. This species also produces large quantities of pollen and can be a significant localized source of allergen.



Biology of reed canarygrass

Reed canarygrass infestation is an important concern in the design and construction of wetland and riparian creation, enhancement, or restoration projects. Infestations may be present on a project site, or reed canarygrass may become established after construction of the project. In such cases weed management measures have included mowing, herbicide application, grazing, cultivation, burning, micronutrient management (boron), macronutrient management (nitrogen), shading (competitive exclusion), flooding, mechanical barriers, and bombing (Naglich, 1994). The growth and reproduction of reed canarygrass is important in understanding the efficacy of these measures.

Sexual reproduction

Reed canarygrass establishes on a site either via seed or by the arrival of rhizome pieces or rootwads. Seeds have the ability to germinate immediately upon ripening and have no known dormancy requirement (Apfelbaum and Sams 1987). No information is available on the longevity of viable seed. A seedbank study in Illinois showed that the seedbank in a reed canarygrass stand was often completely dominated by this species (Apfelbaum and Sams 1987). Although typically fertile and showing good germination, seed appears to have some characteristic that impedes rapid stand development, at least in western Washington (Fransen 1996, Wasser 1982). In fact, a field may require several consecutive seedings for acceptable agricultural stand development in that area (Fransen 1996).

There is some speculation that seedlings do not compete well with seedlings of nonjointed grasses, particularly such creeping grasses as redtop (*Agrostis alba*) and creeping red fescue (*Festuca rubra*) or nongrass species, such as sedges (*Carex* spp.) and rushes (*Juncus* spp.). Reed canarygrass grows vertically for 5 to 7 weeks after germination, after which tillering occurs (Comes et al. 1981). This delay in tillering may limit its early competitive ability in the face of rapidly tillering species, such as redtop and creeping red fescue. Once tillering begins, however, reed canarygrass gains distinct competitive advantage. NRCS is currently testing reed canarygrass establishment in the presence of mixtures of native sedges, annual ryegrass, and native wetland grasses (USDA NRCS 1996).

Vegetative growth and phenology

In the Pacific Northwest, reed canarygrass is one of the earliest grasses to begin growth in late winter and early spring. Growth may start as early as mid-December in some years, with foliage potentially reaching 0.5 meter by mid-March. Such growth is initiated and developed largely at the expense of previously established food reserves in the underground stem system.

Reed canarygrass forms dense, highly productive monocultures that spread radially from that rhizome system. Though rhizomes are thick and densely produced, this species is relatively shallow-rooted. In one study, at least 88 percent of the new shoots in a stand originated from the upper 5 centimeters of soil; 100 percent of shoots arose from the upper 20 centimeters of soil (Comes et al. 1981). Laboratory studies have shown that 74 percent of new shoots originate from rhizomes. Remaining shoots arise from axillary buds on basal nodes (Casler and Hovin 1980). Vegetative vigor is related to maximum root and shoot production. Significantly increased growth was found to be associated with nitrogen enrichment (Decker et al. 1967).

Productivity of reed canarygrass in Wisconsin peaked between mid-June (just before seed ripening) and mid-August, with aboveground standing crop greatest in September (Klopatek and Stearns 1978). Plants continue producing new shoots until the occurrence of hard frosts, usually sometime in November in the Pacific Northwest, but can also be winter-evergreen in some or most years at low elevations.

In West Virginia, available carbohydrate reserves were found to be greatest during winter months, decreasing to a low point in mid-July, and with conspicuous depletion events occurring as the main apical meristem was elevated (late May) and as the seed head developed (early June) (Decker et al. 1967). Plants accumulated higher concentrations of available carbohydrates in the roots/rhizomes during the fall than at any other time of the year.

Graber et al. (1927) and Forman et al. (2000) found that the quantity, quality, and availability of carbohydrate reserves sharply limited the amount of shoot and root growth. Progressive exhaustion of underground carbohydrate reserves by early, frequent, and complete shoot removal ultimately resulted in reduced biomass production or in the death of plants. Nevertheless, early growth and the large number of growth apices involved give reed canarygrass substantial competitive advantage. This particular aspect of the plant's biology has critical implications in the weed management of this species.

Managing reed canarygrass stands

Management of existing reed canarygrass stands typically involves one of two objectives: reducing the vigor of existing stands or killing stands or portions of stands. Methods that may achieve these goals are described below.

Mowing/grazing/disking/burning/flaming/excavation/mulching

Each reed canarygrass stem is sexually reproductive, annually elevating its growing point above the crown of the plant. This characteristic makes reed canarygrass susceptible to cutting damage, especially when cut low (less than 6 cm) during the joint phase of plant growth (when the growing point is elevated during early and mid-spring). Repeated shoot removal stresses plants by preventing them from recovering between shoot disturbance events. Such stress results in reduced vigor, reduced flowering and seed set, and stand thinning. If extremely stressed, stands may thin sufficiently to allow the germination and establishment of other plant species. Continued removal or disruption of the shoot system is thought to eventually eliminate a stand (Forman et al. 2000). Such removal or disruption can be accomplished by repeated mowing, cultivation, burning, flaming, or excavation.

To control reed canarygrass using cultivation, plants should be cut or fields disked or plowed as the plants are coming into flower. This takes advantage of the low underground carbohydrate reserves present at that time. Hovin et al. (1973) found that all stands of reed canarygrass were killed when the stems were cut off just before or at anthesis. Burning and flaming may also be used to kill emerging shoots. Flaming may be more desirable because it does not raise the air quality and safety issues that prescribed burning raises. In addition, flaming can be done at any time of the year and, in most case, without a burn permit from local jurisdictions. Numerous handheld or implement-based flaming devices are available. Repeated burnings or flamings over a year or more may be needed to eradicate or weaken established stands of reed canarygrass. Dead culms/leaves from the previous year's growth should be removed before flaming to minimize fire and smoke hazards and to create better flame access to emerging shoots. Before providing assistance on a burn or recommending a burn, consult the NRCS burn policy located in the General Manual.

Mulching using deep layers of organic material or sheets of opaque plastic, rubber, road felt, or other material eliminates light to the plants, thereby killing the buried or covered plants. The grass is typically cut to within a few centimeters of the ground before the mulch is placed. The mulching materials need to be sufficiently deep or thick and opaque such that light does not pass through and plants are unable to grow through the medium. Plastic sheet barriers tend to quickly deteriorate in the presence of ultraviolet radiation. Sheet barriers need to be firmly anchored to the ground to prevent being uplifted by growth of reed canarygrass. Use of 1- to 2-meter circles or squares of mulch material around newly installed plants is generally considered a successful technique for establishing individual woody plants in a reed canarygrass stand.

Intensive grazing by cows, sheep, and goats shows promise as a management tool (Wilderman 2000). The grass is most palatable in early spring before levels of alkaloids increase and stems become tough. Under continued overgrazed conditions, a reed canarygrass stand typically thins sufficiently to allow the establishment of desirable native species, many of which have inferior palatability. Unfortunately, reed canarygrass often occurs in wetlands where the practice of grazing animals in wetland and riparian habitats raises a different set of water quality and biodiversity concerns, which may not be balanced by benefits of control. These concerns include fecal contamination, erosion, sedimentation, and damage to desirable plant life.

Flooding

Reed canarygrass is tolerant of prolonged soil saturation, prolonged shallow inundation (particularly during the dormant season), and periodic, short-term, deep inundation. This species survives prolonged flooding by producing anoxia-tolerant rhizomes (Brandle 1983). Reed canarygrass is commonly found in areas subjected to periodic shallow flooding during the first half of the growing season. Reed canarygrass is less tolerant of prolonged deep inundation or seasonal fluctuations in hydrology during the middle part of the growing season (Taylor 1993).

Reed canarygrass may only tolerate deep inundation (at least 30 cm of water) for 2 years before it succumbs (USDA NRCS 1996). Stevens and Vanbianchi (1993) report that permanently flooding areas with more than 1.5 meters of water for at least three growing seasons has successfully eliminated reed canarygrass stands. The length of time this species can withstand deep inundation depends on temperature, current, and silt content of the water (Wheaton 1993).

Flooding is most useful in managed wetland systems that have water control systems capable of impounding sufficient water. Beaver introduction into hydrologically unmanaged wetland systems may offer a related tool in managing reed canarygrass. However, dramatic changes in flooding regimes may adversely impact desirable upland and wetland habitats, a fact that needs to be considered if flooding management of reed canarygrass is an option.

Herbicide application

Application of glyphosate has been shown to be an effective tool and is commonly used in managing stands of reed canarygrass. Application timing may be important in how quickly this nonselective, translocated herbicide is moved into the rhizome system, but this must be balanced by the logistical difficulties of spraying this tall-growing grass. Reed canarygrass can reach heights of 2 meters or more, which presents significant difficulties for herbicide application during the middle and late growing season. Fransen (1996) reports that effective herbicide control has been obtained in stands that had been mown at flowering to a height of 1 meter and then sprayed using backpack sprayers. Stands can also be mown to lesser heights and sprayed after plants have produced a subsequent, but shorter crop of foliage.

Spraying should occur when plants have minimum carbohydrate reserves, at and immediately after flowering. However, maximum carbohydrate accumulation in the rhizome system occurs after July, and thus, herbicide translocation would be greatest during this post-flowering period. In Indiana, Comes et al. (1981) found best control when glyphosate was applied at flowering. General observations in western Washington suggest that late summer or early fall applications are effective. Frequently, a second application is needed in mid-spring of the subsequent year to kill remaining plants.

Shading

Planting of coniferous trees can be effective in eliminating reed canarygrass. The conifers need to be planted relatively densely in wide blocks in both the wetland and its adjoining buffers. If the blocks in buffer or wetland habitats are too narrow, side-lighting allows the reed canarygrass infestation to persist in the understory. In some cases, a situation might call for "designed succession" in which faster-growing species, such as willows (*Salix* spp.), red alder (*Alnus rubra*), and/or cottonwood (*Populus balsamifera* ssp. *trichocarpa*), are planted first. These initial plantings are selectively thinned several years later and underplanted with selected conifer species, which would be expected to establish in the less harsh environment provided by a thinned deciduous canopy. Other methods of reed canarygrass control may need to be used on a short-term basis to ensure the planted conifers establish and overtop the reed canarygrass stand.

Integrated strategies

Combinations of the methods described in this section have been shown effective in reducing stand size and vigor and in some cases in eradicating the grass from an area. Reed canarygrass management at the Ridgefield National Wildlife Refuge in Washington showed the best control was achieved using spring herbicide application and fall disking (Crockett et al. 1995, Kilbride and Paveglio 1999).

An integrated approach showing promise in western Washington combines herbicide use with shading. Herbicides are typically used to eliminate reed canarygrass from large-diameter blocks or circles. Once the grass is dead, the blocks or circles are densely planted with desirable native vegetation, such as willows, appropriate conifers, and/or deciduous shrubs. The planted species typically establish well in the absence of reed canarygrass. As planted areas of dense vegetation grow, their canopy begins to reduce the vigor and cover of adjacent areas of reed canarygrass, largely due to shading. As shaded reed canarygrass areas decline in vigor and density, desirable native plants become established and the planted zones enlarge.

Recommendations

Based on the current knowledge of reed canarygrass biology and ecology and recent efforts in managing this species, several options are available to deal with infestations of reed canarygrass:

- Plant wetland and riparian zones and wetlands with selected tree species, emphasizing conifers. Conifer forests or dense deciduous plantings are effective in eliminating reed canarygrass.
- Inundation with at least 0.3 meter of water for most of the year is effective in reducing the vigor and cover of reed canarygrass. Such inundation is best accomplished by using flow control structures rather than by excavation, which wastes valuable topsoil and is generally expensive. Excavation should be driven by goals for altering hydrologic regimes, rather than as a direct weed management tool.
- If large-scale field-burning is not possible for damaging shoots, consider removing thatch and then flaming.
- Consider herbicide applications as a tool in managing reed canarygrass. Despite known and unknown, short- and long-term effects of herbicides in aquatic systems, glyphosate shows utility in managing reed canarygrass particularly when integrated with other management tools. Short-term adverse effects of aquatic herbicides need to be balanced by considering that aquatic systems may experience significant long-term benefit from such herbicide use.
- Attempt eradication for reed canarygrass infestations on sites to be used for newly constructed wetland/riparian projects. Numerous methods are available for eradicating this species from a project site, including mowing, burning, flaming, disking/harrowing, herbicide applications, and various combinations thereof. Sufficient time should be allowed for repeat treatments to ensure eradication of existing stands. If a project area is too large for eradication management or if funds are limited, localized mulching barriers may be used to create planting spaces for trees and other desirable vegetation.

- When time, topography, and other circumstances allow, deplete the reed canarygrass seed store in existing soils prior to restoration plantings. Seed store depletion is accomplished by leaving a cleared project area devoid of vegetation (fallow) for at least one growing season. Multiple fallow growing seasons are preferable to one. Emerging seedlings are killed by repeated disking or harrowing, flaming, and/or periodic herbicide applications. The goal of fallow cultivation is to prevent emerging seedlings or sprouts from tillering and flowering. Repeated cultivations continue to expose new seeds in the soil. Herbicide applications alone deplete seed only in upper elevations of the soil column.
- If seed store depletion is not possible, consider competitive exclusion as a means to discourage reed canarygrass seedling establishment on a cleared/cultivated site. In some situations, seeding competitive grass species, such as tufted hairgrass (*Deschampsia cespitosa*), slough grass (*Beckmannia syzigachne*), bentgrass (*Agrostis* spp.), or turf-forming varieties of red fescue (*Festuca rubra*), may present a significant obstacle to reed canarygrass seedling establishment. Seedings should be heavy (56–112 kg/ha). If used, woody material should be planted before seeding.
- Because much work with reed canarygrass is done in small-scale, nonexperimental situations, most of the information obtained from these efforts is not widely distributed to the larger restoration and scientific communities. In addition, the magnitude and persistence of reed canarygrass infestations must force us to consider this problem in the context of a broad landscape perspective rather than on a site-by-site basis. As a result, an information clearinghouse is needed where interested parties can share their management experiences with this weed or seek information resulting from others' experiences. The Reed Canarygrass Working Group of the Northwest Chapter of the Society for Ecological Restoration is presently serving in this capacity. Contact the author if you wish to connect with this effort.

Literature cited

Apfelbaum, S.I., and C. Sams. 1987. Ecology and control of reed canarygrass. Natural Areas Journal 7:69–74.

Brandle, R. 1983. Evolution der Garungskapazitat in den Flut- und Anoxiatoleranten Rhizomen von *Phalaris arundinacea*, *Phragmites communis*, *Schoenoplectus lacustris*, und *Typha latifolia*. Boticancia Helvetica 93: 39–45.

Casler, M.D., and A.W. Hovin. 1980. Genetics of Vegetative Stand Establishment Characteristics in Reed Canarygrass Clones. Crop Science 20:511–515.

Comes, R.D., L.Y. Marquis, and A.D. Kelley. 1981. Response of seedlings of three perennial grasses to Dalapon, Amitrol, and Glyphosate. Weed Science 29:619-621.

Crockett, R.P., F.L. Paveglio, K.M. Kilbride, and R.B. Wiseman. 1995. Integrated pest management strategies for reed canarygrass (*Phalaris arundinacea* L.) in seasonal wetlands. Abstract, Soc. for Ecol. Restoration 1995 Conf., Seattle, Washington.

Daubenmire, Rexford. 1968. Plant communities. Harper and Row, New York, New York, pp. 238–239.

Decker, A.M., G.A. Jung, J.B. Washko, D.D. Wolf, and M.J. Wright. 1967. Management and productivity of perennial grasses in the Northeast: I. reed canarygrass. West Virginia Univ. Agric. Exp. Sta. Bul. 550T.

Forman, D.J., L. Hardesty, and R.D. Sayler. 2000. Effects Of shade and defoliation on reed canarygrass (*Phalaris arundinacea* L.) biomass production: A greenhouse study. Proc. (abstracts), Reed Canarygrass Conf., Olympia, Washington, p. 6.

Fransen, S. 1996. Agronomist, forages and pastures. Washington State Univ. Coop. Ext., Puyallup. Personal communication to Clayton Antieau, agent for water quality and horticulture, Washington State Univ. Coop. Ext., Jefferson County, Washington.

Galatowitsch, S.M., N.O. Anderson, and P.D. Ascher. 1999. Invasiveness in wetland plants in temperate North America. Wetlands 19(4):733–755.

Graber, L.F., N.T. Nelson, W.A. Luekel, and W.B. Albert. 1927. Organic food reserves in relation to the growth of alfalfa and other perennial herbaceous plants. Wisconsin Agric. Res. Bul. 80.

Guard, B.J. 1995. Wetland plants of Oregon and Washington. Lone Pine Press, Renton, Washington.

Hovin, A.W., B.E. Beck, and G.C. Marten. 1973. Propagation of reed canarygrass from culm segments. Crop Science 13: 747–749.

Kilbride, K.M., and F.L. Paveglio. 1999. Integrated pest management to control reed canarygrass in seasonal wetlands of southwestern Washington. Wildlife Soc. Bul. 27(2):292–297.

Klopatek, J.M., and F.W. Stearns. 1978. Primary productivity of emergent macrophytes in a Wisconsin freshwater marsh ecosystem. American Midland Naturalist 100:320–332.

Merigliano, M.F., and P. Lesica. 1998. The native status of reed canarygrass (*Phalaris arundinacea* L.) in the inland Northwest, USA. Natural Areas Journal 18(3):223–230.

Naglich, F. 1994. Reed canarygrass (*Phalaris arundinacea* L.) in the Pacific Northwest: growth parameters, economic uses, and control. Essay, Master of Environ. Stud., Evergreen State Col., Olympia, Washington.

Stevens, M., and R. Vanbianchi. 1993. Restoring wetlands in Washington. Washington State Dep. Ecol. Pub. 93-17, Olympia, Washington.

Taylor, B.L. 1993. The influence of wetland and watershed morphological characteristics on wetland hydrology and relationships to wetland vegetation communities. M.S.C.E. thesis, Univ. Washington, Seattle, Washington.

United States Department of Agriculture, Natural Resources Conservation Service. 1996. Reed canarygrass control project plan (draft). Plant Materials Centers, Corvallis, Oregon, and Pullman, Washington.

Wasser, C.H. 1982. Ecology and culture of selected species useful in revegetating disturbed lands in the West. U.S. Fish and Wildlife Service, pp. 86–87.

Wheaton, H. 1993. Reed canarygrass, ryegrass, and garrison creeping foxtail. Agric. Pub. G04694, Univ. Missouri, Columbia, Missouri.

Wilderman, D. 2000. Livestock grazing for reed canarygrass management at Trout Lake Natural Area Preserve, Washington. Proc. (abstracts) from Reed Canarygrass Conf., Olympia, Washington, p. 7.

III.J.2.e Control and management of common reed (Phragmites australis (Cav.) Trin. ex Steud.) (PHAU7)

(Marianne Marks, The Nature Conservancy, Arlington, Virginia, author; Beth Lapin and John Randall, The Nature Conservancy, Arlington, Virginia, updated December 1993 and revised 1999; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001. Distribution map and photographs, NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

This abstract provides information on the biology and control of invasive plant species in wetlands.

To the user

Element Stewardship Abstracts (ESAs) are prepared to provide The Nature Conservancy's Stewardship staff and other land managers with management-related information on those species and communities that are most important to protect, or most important to control. The abstracts organize and summarize data from numerous sources including literature and researchers and managers actively working with the species or community.

We hope, by providing this abstract free of charge, to encourage users to contribute their information to the abstract. This sharing of information will benefit all land managers by ensuring the availability of an abstract that contains up-to-date information on management techniques and knowledgeable contacts. Contributors of information will be acknowledged within the abstract and receive updated editions. To contribute information, contact the editor whose address is listed above.

For ease of update and retrievability, the abstracts are stored on computer at the national office of The Nature Conservancy. This abstract is a compilation of available information and is not an endorsement of particular practices or products.

Please do not remove this cover statement from the attached abstract.

1815 North Lynn Street, Arlington, Virginia 22209 (703) 841 5300

Contents

The Nature Conservancy
Element Stewardship Abstract
Common Reed (*Phragmites australis*)

Identifiers

Common name: Common reed Global rank: G5

General description

Phragmites australis is a large perennial rhizomatous grass, or reed. The name *Phragmites* is derived from the Greek word for fence, *phragma*, in reference to its fence-like growth along streams.

Diagnostic characteristics

Members of the genus *Phragmites* are superficially similar to Arundo. Sterile specimens of *P. australis* are sometimes misidentified as *Arundo donax*, a grass introduced to North America from Asia and now troublesome in natural areas, especially in California. The genera can be distinguished when in flower because the glumes of *Phragmites* are glabrous while those of Arundo are covered with soft, whitish hairs 6-8 mm long. In addition, the glumes are much shorter than the lemmas in *Phragmites*.

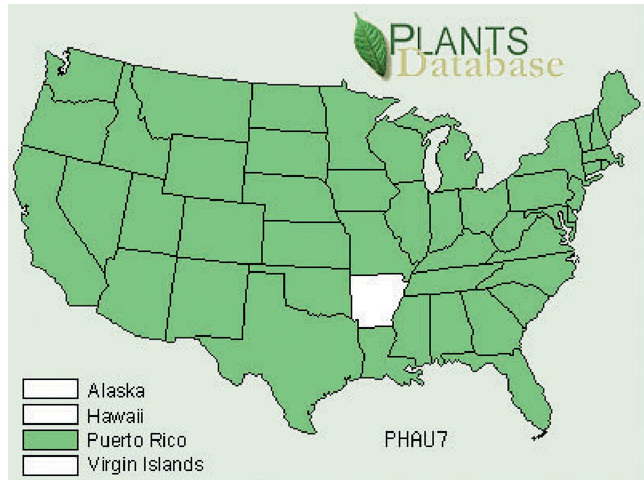
Stewardship summary

Communities that have stable *Phragmites* populations present, but have been exposed to disturbance should be closely monitored. Management is necessary when evidence indicates that *Phragmites* has spread or is spreading and threatening the integrity of rare communities, invading the habitat of rare plants or animals, or interfering with the wildlife support function of refuges. Cutting, burning, application of herbicides (in particular Rodeo™), or water management schemes are possible control measures. The measure(s) used depends on a number of factors including the size and location of the infestation, the presence of sensitive rare species, and the workforce available.

Natural history

Range

Phragmites australis is found on every continent except Antarctica and may have the widest distribution of any flowering plant (Tucker 1990). It is common in and near freshwater, brackish and alkaline wetlands in the temperate zones worldwide. It may also be in some tropical wetlands, but is absent from the Amazon Basin and Central Africa. It is widespread in the United States, typically growing in marshes, swamps, fens, and prairie potholes, usually inhabiting the marsh-upland interface where it may form continuous belts (Roman et al. 1984).



Because *Phragmites* has invaded and formed near-monotypic stands in some North American wetlands only in recent decades, there has been some debate as to whether it is indigenous to this continent or not. Convincing evidence that it was here long before European contact is now available from at least two sources. Niering and Warren (1977) found remains of *Phragmites* in cores of 3,000-year-old peat from tidal marshes in Connecticut. Identifiable *Phragmites* remains dating from 600 to 900 A.D. and constituting parts of a twined mat and other woven objects were found during archaeological investigations of Anasazi sites in southwestern Colorado (Breternitz et al. 1986).

Although the species itself is indigenous to North America, some suspicion is that new, more invasive genotypes were introduced from the Old World (Metzler and Rozsa 1987). Hauber et al. (1991) found that invasive *Phragmites* populations in the Mississippi River Delta differed genetically from a more stable population near New Orleans. They also examined populations elsewhere on the gulf coast, from extreme southern Texas to the Florida panhandle, and found no genetic differences between those populations and the one near New Orleans (Hauber, pers. comm. 1992). This increased their suspicion that the invasive biotypes were introduced to the Delta from somewhere outside the gulf relatively recently.

Phragmites is frequently regarded as an aggressive, unwanted invader in the East and Upper Midwest. It has also earned this reputation in the Mississippi River Delta of southern Louisiana, where over the last 50 years, it has displaced species that provided valuable forage for wildlife, particularly migratory waterfowl (Hauber et al. 1991). In other parts of coastal Louisiana, however, it is feared that *Phragmites* is declining as a result of increasing saltwater intrusion in the brackish marshes it occupies. *Phragmites* is apparently decreasing in Texas as well due to invasion of its habitat by the alien grass *Arundo donax* (Poole, pers. comm. 1985). Similarly, *Phragmites* is present in the Pacific States, but is not regarded as a problem there. In fact, throughout the Western United States there is some concern over decreases in the species habitat and losses of populations.

Habitat

Phragmites is especially common in alkaline and brackish (slightly saline) environments (Haslam 1972, 1971b) and can thrive in highly acidic wetlands (Rawinski, pers. comm. 1985). However, *Phragmites* does not require, nor even prefer these habitats to freshwater areas. Its growth is greater in fresh water, but it may be outcompeted in these areas by other species that cannot tolerate brackish, alkaline, or acidic water. It is often found in association with other wetland plants including species from the following genera: *Spartina*, *Carex*, *Nymphaea*, *Typha*, *Glyceria*, *Juncus*, *Myrica*, *Triglochin*, *Calamagrostis*, *Galium*, and *Phalaris* (Howard et al. 1978).

Phragmites occurs in disturbed areas as well as pristine sites. It is especially common along railroad tracks, roadside ditches, and piles of dredge spoil, wherever even slight depressions hold water (Ricciuti 1983). Penko (pers. comm. 1993) has observed stunted *Phragmites* growing on acidic tailings (pH 2.9) from an abandoned copper mine in Vermont. Various types of human manipulation and/or disturbance are thought to promote *Phragmites* (Roman et al. 1984). For example, restriction of the tidal inundation of a marsh may result in lowering of the water table, which may in turn favor *Phragmites*. Likewise, sedimentation may promote the spread of *Phragmites* by elevating a marsh's substrate surface and effectively reducing the frequency of tidal inundation (Klockner, pers. comm. 1985).

A number of explanations have been proposed to account for the recent dramatic increases in *Phragmites* populations in the Northeastern and Great Lakes States. As noted above, habitat manipulations and disturbances caused by humans are thought to have a role. In some areas *Phragmites* may also have been promoted by the increases in soil salinity that result when de-icing salt washes off roads and into nearby ditches and wetlands (McNabb and Batterson 1991). On the other hand, bare patches of road sand washed into ditches and wetlands may be of greater importance. *Phragmites* seeds are shed from November through January and so may be among the first propagules to reach these sites. If the seeds germinate and become established, the young plants usually persist for at least 2 years in a small, rather inconspicuous stage, resembling many other grasses. Later, perhaps after the input of nutrients, they may take off and assume the tall growth form that makes the species easily identifiable. Increases in soil nutrient concentrations may come from runoff from farms and urban areas. It has also been suggested increases in nutrient concentrations, especially nitrates, are primarily responsible for increases in *Phragmites* populations. Ironically, eutrophication and increases in nitrate levels are sometimes blamed for the decline of *Phragmites* populations in Europe (Den et al. 1989).

Ecology

Salinity and depth to the water table are among the factors that control the distribution and performance of *Phragmites*. Maximum salinity tolerances vary from population to population; reported maxima range from 12 ppt (1.2%) in Britain to 29 ppt in New York to 40 ppt on the Red Sea coast (Hocking et al. 1983). Dense stands normally lose more water through evapotranspiration than is supplied by rain (Haslam 1970). However, rhizomes can reach down almost 6 feet below ground, their roots penetrating even deeper, allowing the plant to reach low-lying ground water. Killing frosts may knock the plants back temporarily, but can ultimately increase stand densities by stimulating bud development (Haslam 1968).

Phragmites has a low tolerance for wave and current action that can break its culms (vertical stems) and impede bud formation in the rhizomes (Haslam 1970). It can survive, and in fact thrive, in stagnant water where the sediment is poorly aerated at best (Haslam 1970). Air spaces in the aboveground stems and in the rhizomes themselves assure the underground parts of the plant with a relatively fresh supply of air. This characteristic and the species' salinity tolerance allow it to grow where few others can survive (Haslam 1970). In addition, the buildup of litter from the aerial shoots within stands prevents or discourages other species from germinating and becoming established (Haslam 1971a). The rhizomes and adventitious roots themselves form dense mats that further discourage competitors. These characteristics are what enable *Phragmites* to spread, push other species out, and form monotypic stands.

Such stands may alter the wetlands they colonize, eliminating habitat for valued animal species. On the other hand, the abundant cover of litter in *Phragmites* stands may provide habitat for some small mammals, insects, and reptiles. The aerial stems provide nesting sites for several species of birds, and song sparrows have been seen eating *Phragmites*' seeds (Klockner, pers. comm. 1985). Muskrats (*Ondatra zibethicus*) use *Phragmites* for emergency cover when low-lying marshes are swept by storm tides and for food when better habitats are overpopulated (Lynch et al. 1947).

Studies conducted in Europe indicate that gall-forming and stem-boring insects may significantly reduce growth of *Phragmites* (Durska 1970; Pokorny 1971). Skuhravy (1978) estimated that roughly a third of the stems in a stand might be damaged, reducing stand productivity by 10 to 20 percent. Mook and van der Toorn (1982) found yields were reduced by 25 to 60 percent in stands heavily infested with lepidopteran stem- or rhizome-borers. Hayden (1947) suggested that aphids (*Hyalopterus pruni*) heavily damaged a *Phragmites* stand in Iowa. On the other hand, Pintera (1971) indicated that although high densities of aphids may bring about reductions in *Phragmites* shoot height and leaf area, they had little effect on shoot weight. Like other emergent macrophytes, *Phragmites* has tough leaves and appears to suffer little grazing by leaf-chewing insects (Penko 1985).

As mentioned, there is great concern about recent declines in *Phragmites* in Europe where the species is still used for thatch. In fact, the journal Aquatic Botany devoted an entire issue (vol. 35, no.1, September 1989) to this subject. Factors believed responsible for the declines include habitat destruction and manipulation of hydrologic regimes by humans, grazing, sedimentation, and decreased water quality (eutrophication) (Ostendorp 1989).

Detailed reviews of the ecology and physiological ecology of *Phragmites* are provided by Haslam (1972) and Hocking et al. (1983) and an extensive bibliography is provided by van der Merff et al. (1987).

Reproduction

Phragmites is typically the dominant species in areas that it occupies. It is capable of vigorous vegetative reproduction and often forms dense, virtually monospecific stands. Hara et al. (1993) classify sparse stands as those with densities of less than 120 culms per square yard and dense stands as those with densities of up to about 240 culms per square yard in wet areas or up to 360 culms per square yard in dry areas. Mammalian and avian numbers and diversity in the dense stands are typically low (Jones and Lehman 1987). Newly opened sites may be colonized by seed or by rhizome fragments carried to the area by humans in soils and on machinery during construction or naturally in floodwater.

The plants generally flower and set seed between July and September and may produce great quantities of seed. In the Northeast, seeds are dispersed between November and January. However, in some cases most or all of the seed produced is not viable (Tucker 1990). The seeds are normally dispersed by wind, but may be transported by birds, such as redwinged blackbirds that nest among the reeds (Haslam 1972). Following seed set, nutrients are translocated down into the rhizomes, and the aboveground portions of the plant die back for the season (Haslam 1968).

Temperature, salinity, and water levels affect seed germination. Water depths of more than 5 centimeters and salinity above 20 ppt (2%) prevent germination (Kim et al. 1985, Tucker 1990). Germination is not affected by salinity below 10 ppt (1%), but declines at a higher salinity. Percentage germination increases with increasing temperature from 16 to 25 degrees Celsius, while the time required to germinate decreases from 25 to 10 days over the same temperature range. Barry Truitt (pers. comm. 1992) observed that areas covered by thick mats of wrack washed up during storms and high water events are frequently colonized by *Phragmites* on the Virginia Coast Reserve. It is not clear whether it establishes from rhizome pieces washed in with the wrack or from seed that blows in later.

Once a new stand of *Phragmites* takes hold, it spreads predominantly through vegetative reproduction. Individual rhizomes live for 3 to 6 years, and buds develop at the base of the vertical type late in the summer each year. These buds mature and typically grow about 3 feet (up to 33 feet in newly colonized, nutrient-rich areas) horizontally before terminating in an upward apex and going dormant until spring. The apex then grows upward into a vertical rhizome, which in turn produces buds that will form more vertical rhizomes. Vertical rhizomes also produce horizontal rhizome buds, completing the vegetative cycle. These rhizomes provide the plant with a large, absorbent surface that brings the plant nutrients from the aquatic medium (Chuzhoza and Arbuzova 1970). The aerial shoots arise from the rhizomes. They are most vigorous at the periphery of a stand where they arise from horizontal rhizomes, as opposed to old verticals (Haslam 1972).

Condition

Threats

Impacts (threats posed by this species)—
Phragmites can be regarded as a stable, natural component of a wetland community if the habitat is pristine and the population does not appear to be expanding. Many native populations of *Phragmites* are "benign" and pose little or no threat to other species. These populations should be left intact. Examples of areas with stable, native populations include sea-level fens in Delaware and Virginia and along Mattagota Stream in Maine (Rawinski 1985, pers. comm. 1992). In Europe, a healthy reed belt is defined as a "homogeneous, dense or sparse stand with no gaps in its inner parts, with an evenly formed lakeside borderline without aisles, shaping a uniform fringe or large lobes, stalk length decreasing gradually at the lakeside border, but all stalks of one stand of similar height; at the landside edge the reeds are replaced by sedge or woodland communities or by unfertilized grasslands" (Ostendorp 1989).

Stable populations may be difficult to distinguish from invasive populations, but such factors as site disturbance and the earliest collection dates of the species should be examined to arrive at a determination. If available, old and recent aerial photos can be compared to determine whether stands in a given area are expanding (Klockner, pers. comm. 1985).

Phragmites is a problem when and where stands appear to be spreading while other species typical of the community are diminishing. Disturbances or stresses, such as pollution, alteration of the natural hydrologic regime, dredging, and increased sedimentation, favor invasion and continued spread of *Phragmites* (Roman et al. 1984). Other factors that may have favored recent invasion and spread of *Phragmites* include increases in soil salinity (from fresh to brackish) and/or nutrient concentrations, especially nitrate, and the introduction of a more invasive genotype(s) from the Old World (McNabb and Batterson 1991; Metzler and Rozsa 1987, see Global range section for further information).

Michael Lefor asserts that one reason for the general spread of *Phragmites* has been the destabilization of the landscape (pers. comm. 1993). In urban landscapes water is apt to collect in larger volumes and pass through more quickly (flashily) than formerly. This tends to destabilize substrates leaving bare soil open for colonization. Watersheds throughout Eastern North America are flashier because of the proliferation of paved surfaces, lawns, and roofs, and the fact that upstream wetlands are largely filled with post settlement/post agricultural sediment from initial landclearing operations.

Many Atlantic coast wetland systems have been invaded by *Phragmites* as a result of tidal restrictions imposed by roads, water impoundments, dikes, and tide gates. Tide gates are installed to drain marshes to harvest salt hay, to control mosquito breeding, and, most recently, to protect coastal development from flooding during storms. This alteration of marsh systems may favor *Phragmites* invasion by reducing tidal action and soil-water salinity and lowering water tables.

Phragmites invasions may threaten wildlife because they alter the structure and function (wildlife support) of relatively diverse Spartina marshes (Roman et al. 1984). This is a problem on many of the eastern coastal National Fish and Wildlife Refuges including Brigantine in New Jersey; Prime Hook and Bombay Hook in Delaware; Tinicum in Pennsylvania; Chincoteague in Virginia; and Trustom Pond in Rhode Island.

Plant species and communities threatened by *Phragmites* are listed in the Monitoring section. Some of these instances are described below:

- **Massachusetts**—a brackish pondlet near Horseneck Beach supports the State rare plant *Myriophyllum pinnatum* (Walter) BSP, which *Phragmites* is threatening by reducing the available open water and shading aquatic vegetation (Sorrie, pers. comm. 1985).
- **Maryland**—at Nassawango Creek, a rare Coastal Plain peatland community is threatened by *Phragmites* (Klockner, pers. comm. 1985).
- **Ohio**—at the Arcola Creek wetland, *Phragmites* is threatening the State endangered plant *Carex aquatilis* Wahlenb (Young, pers. comm. 1985).

Phragmites invasions also increase the potential for marsh fires during the winter when the aboveground portions of the plant die and dry out (Reimer 1973). Dense congregations of redwing blackbirds, which nest in *Phragmites* stands preferentially, increase chances of airplane accidents nearby. The monitoring and control of mosquito breeding is nearly impossible in dense *Phragmites* stands (Hellings and Gallagher 1992). In addition, *Phragmites* invasions can have adverse aesthetic impacts. In Boston's Back Bay Fens, dense stands have obscured vistas intended by the park's designer, Frederick Law Olmstead (Penko, pers. comm. 1993).

As noted, *Phragmites* is not considered a threat in the West or most areas in the Gulf States.

Restoration potential

Areas that have been invaded by *Phragmites* have excellent potential for recovery. Management programs have proven that *Phragmites* can be controlled, and natural vegetation will return. However, monitoring is imperative because *Phragmites* tends to reinvade. Control techniques may need to be applied several times or, perhaps, in perpetuity. Some areas have been so heavily manipulated and degraded that *Phragmites* may be impossible to eliminate from them. For example, it may be especially difficult to control *Phragmites* in freshwater impoundments that were previously salt marshes.

Management/monitoring

Management requirements

Invasive populations of *Phragmites* must be managed to protect rare plants that it might outcompete, valued animals whose habitat it might dominate and degrade, and healthy ecosystems that it might greatly alter.

Management programs

Cultural, mechanical, and/or chemical methods can be used to control *Phragmites.* The factors that are believed responsible for the alarming decreases of *Phragmites* beds in Europe and Texas—habitat destruction, increased soil nitrate levels, and eutrophication (Boar, Crook, and Moss 1989, Ostendorp 1989, Sukopp and Markstein 1989)—are not appropriate as management tools in natural areas.

***Biological control*—**Biological control does not appear to be an option at this time. No organisms that significantly damage *Phragmites australis*, but do not feed on other plant species have been identified. Naturally occurring parasites have not proven to be successful controls (Tscharntke 1988, Mook and van der Toorn 1982, van der Toorn and Mook 1982). In addition, some of the arthropods that feed on *Phragmites* are killed by winter fires and thus would most likely be eliminated from the systems where prescribed fires are used. Coots, nutria, and muskrats may feed on *Phragmites*, but appear to have limited impacts on its populations (Cross and Fleming 1989).

***Burning*—**Prescribed burning does not reduce the growing ability of *Phragmites* unless root burn occurs. Root burn seldom occurs, however, because the rhizomes are usually covered by a layer of soil, mud, and/or water. Fires in *Phragmites* stands are dangerous because this species can cause spot fires over 100 feet away (Beall 1984). Burning does remove accumulated *Phragmites* leaf litter, giving the seeds of other species area to germinate. Prescribed burning has been used with success after chemical treatment for this purpose at The Brigantine National Wildlife Refuge, NJ (Beall 1984) and in Delaware (Lehman, pers. comm. 1992). Occasional burning has been used in Delaware in conjunction with intensive spraying and water level management. This helps remove old canes and allows other vegetation to grow (Daly, pers. comm. 1991). **Before providing assistance on a burn or recommending a burn, consult the NRCS burn policy in the General Manual.**

At Wallops Island, Virginia, a small (100 by 400 foot) brackish to saline to dry wetland was burned November 1990 to control *Phragmites* (M. Ailes, pers. comm. 1992). A variety of other species appeared in the year following the burn, but they appeared leggy while the *Phragmites* remained vigorous.

At Wertheim National Wildlife Refuge in New York, a 20- to 30-acre freshwater impoundment was drained in the fall of 1989, burned the following winter, and then reflooded (Parris, pers. comm. 1991). *Phragmites* was eliminated from the half of the marsh that was treated, and the area remained free of the grass through 1992.

According to Cross and Fleming (1989), late summer burns may be effective, but winter and spring burning may in fact increase the densities of spring crops. Thompson and Shay (1985) performed experimental burn treatments on Delta Marsh, Manitoba. They found that spring, summer, and fall burns resulted in higher total shoot densities and lower mean shoot weights than on controls primarily as a result of greater densities of shorter, thinner vegetative shoots. Shoot biomass was greater in spring-burned and fall-burned plots than in control areas, but less on summer-burned plots. They also found that belowground production increased following spring and fall burns, but not following summer burns. The increase in light availability following burns generally appears to benefit *Phragmites.* A variety of understory responses to these burns was noted. For example, summer burns increased species diversity, richness, and evenness, although certain species declined.

In Connecticut, a late spring burn followed by manual flooding with salt water was successful in reducing *Phragmites* height and density (Steinke, pers. comm. 1992). After 3 years, the fuel load was exhausted. The process was expensive, and self-regulating tide gates were installed instead (see Manipulation of water level and salinity).

In Europe, experimental removal of litter in winter resulted in doubling the aboveground biomass (Graneli 1989). Increased light availability at the soil surface and aeration of the soil around the rhizomes may have been responsible for this increase. Burning in the winter in an experimental field caused little damage, while burning during the emergence period led to the death of the majority of *Phragmites* shoots (van der Toorn and Mook 1982).

Chemical—Rodeo™, a water solution of the isopropylamine salt of glyphosate, is commonly used for *Phragmites* control. This herbicide is not, however, selective and kills grasses and broadleaved plants alike. Toxicity tests indicate that it is virtually nontoxic to all aquatic animals tested. It should be noted that many of these tests were performed by or for Monsanto, the company that manufactures Rodeo™. Bioconcentration values for glyphosate in fish tissues were insignificant. Glyphosate biodegrades quickly and completely in the environment into natural products including carbon dioxide, nitrogen, phosphate, and water. Finally, since glyphosate does not volatilize, it will not vaporize from a treated site and move to a nontarget area (Brandt 1983; Comes, Bruns, and Kelly 1976; Folmar, Sanders, and Julin 1979; Monsanto 1985).

Rodeo™ must be mixed with water and a surfactant that allows it to stick to and subsequently be absorbed by the plant (Beall 1984). Instructions for application, amounts needed per acre, the approved surfactants, and ratios for mixing are on the Rodeo™ label. Glyphosate must be mixed with clean or, if possible, distilled water because it binds tightly to sediment and is thus rendered nontoxic to plants (Lefor, pers. comm. 1992). This limits its effectiveness, but also may help prevent it from acting on plants that were not originally targeted. Rodeo™ should not by applied in windy conditions as the spray will drift (I. Ailes, pers. comm. 1985). It also should not be applied if rain is forecast within 12 hours because it will wash away before it has a chance to act (Daly 1984). Application rates may vary, but, as one example, effective control of *Phragmites* in a Delaware marsh was achieved with 4 pints per acre of concentrate (Lehman, pers. comm. 1992).

Application of Rodeo™ must take place after the tasseling stage when the plant is supplying nutrients to the rhizome. At this time, when Rodeo™ is sprayed onto the foliage of aquatic weeds, it translocates into the roots. Rodeo™ interferes with essential plant growth processes, causing gradual wilting, yellowing, browning, and deterioration of the plant. Studies on tasseling at the Augustine Tidal area, in Port Penn, Delaware, indicated that tasseling in a stand is never 100 percent, but that it is possible to spray when 94 percent of the plants are tasseling. In dense stands, subdominant plants are protected by the thick canopy and thus may not receive adequate herbicide. For these reasons, touchup work is necessary (Lehman 1984).

At Brigantine National Wildlife Refuge, Rodeo™ was applied aerially after the plants tasseled in late August. The application resulted in a 90 percent success. The following February, a fast-moving prescribed burn was carried out to remove litter, exposing the seedbed for reestablishment of marsh vegetation. However, funding was not available for several years and *Phragmites* has returned to 90 percent of the previously treated areas (Beall, pers. comm. 1991). Treatment was resumed in fall 1991.

In September 1983, at the Prime Hook Wildlife Refuge in Delaware, 500 acres of freshwater impoundments were sprayed with Rodeo™ from a helicopter for *Phragmites* control. The plants yellowed within 10 days. The following May, aerial and ground evaluations of the sprayed area revealed a 98 percent kill of *Phragmites* (Daly 1984). In addition to applying herbicide, Prime Hook manipulates water levels with a stop log to stress *Phragmites;* winter water levels are held at an elevation of 2.8 feet mean sea level until June, when water would otherwise be held at 2.2 feet mean sea level. The combined spraying and water management approach was successful, and many aquatic plants returned. A regime of spraying in August and September for 2 years followed by flooding was used through 1991 (Daly, pers. comm. 1991). Annual costs of *Phragmites* control are $20,000 annual at Prime Hook (1,000 acres) and $3,000 at Bombay Hook (20–60 acres); monitoring costs, which include reading vegetation transects for species presence and density each September, are not included in the cost.

Aerial spraying has been used since 1983 in many Delaware State wildlife refuges (Lehman, pers. comm. 1992). Using Rodeo™, the State sprays freshwater and brackish impoundments, brackish marshes, and salt marshes from early in September to early in October. This is combined with winter burns between the first and second year of spraying. Areas are spot-treated whenever needed after that. The herbicide treatments consist of 4 pints per acre the first year and 2 pints per acre the second, with an average cost of $65 per acre. The State is involved with cost-sharing programs with private landowners where the State pays half the spraying cost with a willing owner. Desirable native vegetation usually returns after spraying; no revegetation is done. Occasionally, the areas become open mud flats that are eventually repopulated by *Phragmites*.

At Chincoteague National Wildlife refuge, an aerial spraying program initiated in 1986 in an 18-mile-long freshwater impoundment was terminated due to budget cuts. *Phragmites* quickly reclaimed the area, estimated to be 100 to 150 acres total in small scattered stands (I. Ailes, pers. comm. 1991). In September 1991, spraying with Rodeo™ began again. Because the area is impounded, the water level usually is lower in the spring, which helps prevent *Phragmites* regrowth.

Herbicides are used at Tinicum Environmental Center because other control options are limited. Unplanned burns do occur, but prescribed burns are not allowed due to the proximity to the highway and airport. Tinicum was granted $2 million to restore an 18-acre site. They altered the elevation of the marsh, seeded with native plants, and monitored the results (Nugent, pers. comm. 1991).

At Parker National Wildlife Refuge, an aerial spraying program (annual budget $5,000) for 50 acres of a 100-acre freshwater impoundment began in mid-August 1991. A winter burn is anticipated and a second year of spraying planned. Results will be monitored by using aerial photos to delineate the boundaries of the *Phragmites* clones. A nearby tower also provides a suitable viewing point to observe progress (Healey, pers. comm. 1992).

In more fragile situations where *Phragmites* is threatening a rare plant or community, aerial spray techniques are inappropriate because such large-scale application could kill the community that the entire operation was designed to protect. Glyphosate can be applied to specific plants and areas by hand with a backpack sprayer. Wayne Klockner of The Nature Conservancy's Maryland Field Office has been successful in eliminating most *Phragmites* at the Nassawango preserve by applying glyphosate by hand with a backpack sprayer (Klockner, pers. comm. 1985). The control program there began in 1983; actual spraying is conducted along the power line right-of-way by Delmarva Power (Droege, pers. comm. 1991). Delmarva Power generally sprays with trucks, backpacks, or helicopter, depending on the accessibility of the area and presence of rare plants nearby (Johnstone, pers. comm. 1991). They use Rodeo™ in tidal areas and Accord™ (another glyphosate product) in nontidal areas from mid-August to mid-October, when the plants are going to seed. They spray intensively the first year, and conduct touchup spraying the second year, which eliminates 90 to 95 percent of the plants. They then return every 3 years to eliminate any new plants. They do not spray if the plants are not tasselling and are short.

Rodeo™ was used at Cape May Meadows in 1989, 1990, and 1991. It was applied with a 30-gallon gas-powered tank with spray nozzle mounted on a truck, Indian pump sprayers, 2.5-gallon handheld sprayers, and wick applicators (Johnson, pers. comm. 1991). This appeared to kill most, if not all, of the treated *Phragmites* in this 20-acre area. Plants found in the area following treatment were shorter, and the stand was less dense (determined visually). However, the dead stalks remained and blocked views from the trail.

In Connecticut, a 5- by 23-meter patch of *Phragmites* was treated with a handheld spray of Rodeo™ (1988 and 1989) and Roundup™ (1990 and 1991) for 4 years in late August to early September. The *Phragmites* is shorter and less dense at the site, but it is still present (Lapin pers. observ.). Actions to supplement and enhance herbicide applications including the removal of tassels (1991) and dead stalks (1992) were taken.

Other chemicals have been used on *Phragmites* and are described in Cross and Fleming (1989).

Also see Cutting at Constitution Marsh for another method of application.

Cutting—Cutting has been used successfully to control *Phragmites*. Since it is a grass, cutting several times during a season at the wrong times may increase stand density (Osterbrock 1984). However, if cut just before the end of July, most of the food reserves produced that season are removed with the aerial portion of the plant, reducing the plant's vigor. This regime may eliminate a colony if carried out annually for several years. Care must be taken to remove cut shoots to prevent their sprouting and forming stolons (Osterbrock 1984). In the Arcola Creek Preserve in Ohio, cutting reduced the vigor of the *Phragmites* colony. Also in Ohio, at Morgan Swamp, cutting began in mid to end of July (before tassel set) in 1989 around a gas well in a freshwater wetland (Seidel, pers. comm. 1991). The preferred tool was an old-fashioned hedge trimmer with an 8-inch flat blade with serrations manufactured by Union Fork and Hoe. The trimmers worked better than loppers and were safer than sickles. A circular blade on a weed whacker was also used and proved to be faster and good for staff, but it was more dangerous for volunteers and detracted from the atmosphere of the workday (Huffman, pers. comm. 1992).

Small patches (10 by 50 feet) in a New York freshwater system were cut at the end of July or the beginning of August for 2 successive years with positive results (Schneider, pers. comm. 1990). The handcut material was removed from the site and thrown on a brush pile. (Unfortunately, it was located too close to the water and returned to the system.)

Massachusetts Audubon staff have cut the perimeter around a 0.25-acre *Phragmites* patch at the end of July since 1986 in a freshwater wetland at Daniel Webster Preserve in Marshfield, Massachusetts (Anderson, pers. comm. 1992). They have monitored their success in keeping it from spreading by using a map and hand compass.

Stands of *Phragmites* of less than 1 acre in extent that block views in Everglades National Park are cut just before the onset of the rainy season. The rise in water elevation from the rains that follow stresses the roots of the plant. This works to a degree, but *Phragmites* returns (Dowlen, pers. comm. 1985).

In Quincy, Massachusetts, the town used small Bobcats with lawnmower clippers mounted on the buckets with a flexible cable to cut an area with 75 percent cover of *Phragmites* and 20 to 25 feet of muck (Wheelwright, pers. comm. 1991; Dobberteen pers. comm. 1991). Cutting this 10-acre plot three times during the summer (April, June, August) cost $150,000. The cut material was stockpiled nearby where it was to be burned in the winter when it was washed away in a severe storm.

Cutting culms to 6 inches followed by addition of rock salt on a 10- by 10-foot patch appeared to have reduced the height and density of *Phragmites* in a salt marsh in Greenwich, Connecticut (Jontos and Allan 1984). Continued observations indicated that this trend appeared to continue (Jontos, pers. comm. 1992).

Cutting an 25- by 25-foot area to waist height with hedge clippers and applying one drop of Roundup™ with a large-needle (horse size) syringe into the top of the plant in a brackish-freshwater marsh was begun in Constitution Marsh in New York in 1991 (Keene, pers. comm. 1991). Initial results indicate 90 percent eradication.

In Connecticut, cutting below the first leaf at the end of July in 1986, 1989, 1990, 1991, and 1992 in a freshwater tidal wetland around the perimeter of a 1-acre patch prevented subsequent expansion of the patch. Monitoring using aerial photos taken at 5-year intervals indicated the control success. Cutting was done with handheld cutters and gas-powered hedge trimmers, which were efficient. Cut material was removed from the site and allowed to decompose in upland areas. In a second area, similar efforts in a calcareous wetland were monitored by placing red survey wires around the perimeter of the patch. Preliminary observations indicate a cessation of *Phragmites* expansion.

In Europe, Weisner and Graneli (1989) found that oxygen transport was reduced by cutting the culms above and below the water surface. Cutting below the water in June almost totally inhibited regrowth of shoots the following summer, while cutting above water reduced regrowth of shoots. Cutting in August did not reduce growth the following summer. Cutting in sandy substrates was minimally effective, while cutting on calcareous muds caused decreases in oxygen levels.

Also see Manipulation of water level and salinity.

Grazing, dredging, and draining—Grazing, dredging, and draining are often used to reduce stand vigor (Howard, Rhodes, and Simmers 1978). However, draining and dredging are not appropriate for use on most preserves (Osterbrock 1984).

Grazing may trample the rhizomes and reduce vigor, but the results are limited (Cross and Fleming 1989). Van Deursen and Drost (1990) found that cattle consumed 67 to 98 percent of aboveground biomass. In a 4-year study, they found that reed populations may reach new equilibria under grazing regimes.

Manipulation of water level and salinity—A self-regulating tidegate that reintroduced saltwater tidal action was used to help restore a diked marsh in

Fairfield, Connecticut (Steinke, pers. comm. 1992; Bongiorno et al. 1984). A 1- to 3-foot reduction in stem height resulted over each of 3 years. In addition to reduced height, plant density declined dramatically from 13.6 plants per square yard in 1980 to 4 plants per square yard the following year. In following years, *Phragmites* continued to decline, although less dramatically. In addition to the decreased height and density of the *Phragmites* stands, typical marsh flora including *Salicornia*, *Distichlis*, *Spartina alterniflora Loisel*, and *S. Patens (Aiton) muhl.* returned. Depending on topography and elevation, *Phragmites* was eliminated in large areas and continued to remain short and sparse in other areas through 1992. Hence, reintroduced tidal action and salinity can reduce *Phragmites* vigor and restore the community's integrity. This has been implemented successfully in other degraded former salt marshes in Connecticut (Rozsa, pers. comm. 1992).

Flooding can be used to control *Phragmites* when 3 feet of water covers the rhizome for an extended period during the growing season, usually 4 months (Beall 1984). However, many areas cannot be flooded to such depths. Furthermore, flooding could destroy the communities or plants targeted for protection.

Open marsh water management (OMWM) has been used to control *Phragmites*. Plugging of ditches and addition of culverts to raise soil salinity appears to have caused *Phragmites* dieback over the last four growing seasons at Fireplace Neck, New York (Niniviaggi, pers. comm. 1991; Rozsa, pers. comm. 1992).

Hellings and Gallagher (1992) found that *Phragmites* was negatively impacted by increasing salinity and increased flooding. They also found that cutting and subsequent flooding reduced growth and survival in outdoor experiments. They suggest that *Phragmites* may be controlled by increasing flooding and salinity levels. Matoh, Matsushita, and Takahashi (1988) also found reduction in vigor with increased salinity. However, death apparently occurred only when cutting was combined with brackish flooding (Hellings and Gallagher 1992).

In Europe, episodic freshwater flooding occurring early in the growing season has been suggested as one of the reasons for reed population declines (Ostendorp 1991). McKee et al. (1989) investigated root metabolic changes due to freshwater flooding, and labeled *Phragmites* as a flood-tolerant species.

Also see Chincoteague National Wildlife refuge under Chemicals, Wertheim National Wildlife refuge under Burning, and Town of Quincy under Cutting for additional references.

Mowing, disking, and pulling—Beall (1984) discourages mowing and disking. Mowing only affects the aboveground portion of the plant, so mowing must occur annually. To remove the rhizome, disking could be employed; however, this could potentially result in an increase of *Phragmites* since pieces of the rhizome can produce new plants. Cross and Fleming (1989) describe successful mowing regimes of several-year duration during the summer (August and September) and disking in summer or fall.

In Cape May Meadows, New Jersey, a brackish to freshwater, nontidal, sandy area, an attempt was made to remove rhizomes by pulling to a depth of 3 feet (Johnson, pers. comm. 1991). This resulted in a sparse *Phragmites* stand the following year. However, it was labor-intensive (using 130 people-hours to cover a 50-ft^2 patch) and could be applied best to sandy soils.

In a private yard, *Phragmites* was mowed and a thin layer of soil and grass seed was added. This was mowed weekly over the course of the summer. In the second summer shoots of *Phragmites* occurred around the edges. The rhizomes were decomposing after this treatment (M. Ailes, pers. comm. 1992).

Plastic—Clear plastic 6 millimeters thick, 12 by 17 meters, weighing 115 pounds, was carried into a North Carolina marsh by air and held in place by sandbags (Boone et al. 1987, 1988). Plants were initially cut to 6 to 8 inches with a hand-pushed bush hog (Boone, pers. comm. 1991) or a weedeater with blade, with a 20- by 20-meter area requiring several days to cut. The cut material was left, and the plastic put over the area. The high temperatures under the plastic caused dieoff of *Phragmites* in 3 to 4 days. After 8 to 10 weeks, the plastic deteriorated. The rhizomes appeared to have died back, but the project was of short duration, and the results were not monitored the following year (Boone, pers. comm. 1991). Turner (pers. comm. 1992) noted that followups in subsequent years indicated *Phragmites* returned, but not as densely. Plastic management in each 12- by 12-meter plot took an average of 53 hours compared with 17 hours to cut and 3 hours to burn (Boone et al. 1987).

Clear plastic in two narrow swaths (70 by 20 meters) was placed along the edge of a tidal brackish pond after handcutting the *Phragmites* at the end of July 1991 (Anderson, pers. comm. 1992). One plot in total sun had a complete kill of *Phragmites* in 10 days, while the plot in partial shade had a partial kill. It is unknown how the plastic was kept in place or what was done with the cut material.

Clear and black plastic were used on 50-foot circular areas at Constitution Marsh in New York in 1990 and 1991 (Keene, pers. comm. 1991). Although there was difficulty because of tidal influence, the plastic was weighted down with rocks and appeared to kill what is under it. Runners along the edge were treated with a syringe application of Roundup™ in August. In November 1991, a hole cut in the middle of the black plastic provided the opportunity for cattail shoots to germinate. After the first year there was viable *Phragmites* in the areas covered. It appeared that the black plastic was more effective because of the higher heat levels attained (Rod, pers. comm. 1992).

Monitoring requirements

Phragmites populations require close monitoring to determine whether they are increasing in area. Populations that are growing may quickly threaten or even eliminate rare elements. Monitoring provides the data needed to decide if control measures are necessary. If and when a control program is begun, targeted populations should be monitored to determine the program's effectiveness. If it is possible to leave untreated control areas without jeopardizing the success of the control program, these areas should be monitored as well for comparison. It is imperative to continue monitoring even if a control program succeeds initially because *Phragmites* may reinvade and the sooner this is detected the easier it will be to combat.

To assess if a *Phragmites* colony is spreading, quantitative measurements should be made of percentage of aerial cover, stem density, and culm height, especially at the periphery of the stand. Annual data should be compared to detect if the colony is expanding and the stand gaining vigor. Inventories of the vegetation in and near the colony should also be carried out to determine whether declines in species diversity are occurring.

In Europe, reed declines have been documented by comparing areas covered by *Phragmites* colonies on up-to-date maps or aerial photographs with older sources, monitoring permanent quadrants within or at the border of the reed belt and mapping the stubble fields left after dieback (Ostendorp 1989). In lakes (Stark and Dienst 1989), wooden poles 5 meters apart were connected with string and the numbers of reed stalks directly below the strings were counted each year in the spring.

Monitoring programs

The programs listed below used various methods to control *Phragmites* populations and are monitoring the success of these actions including the degree of recovery of native species and the longevity of the control.

Connecticut:

- Monitoring *Phragmites* reduction and replacement vegetation after reintroducing tidal flow, using transects and line intercept. Contact: Department of Botany, Connecticut College, New London, CT 06320.
- Monitoring *Phragmites* reaction to reintroduction of tidal flow and salinity. Contact: Fairfield Conservation Commission, Independence Hall, 725 Old Post Road, Fairfield, CT 06430 (203-256-3071).
- Addition of rock salt and casual observation of reduction of *Phragmites* height and density; also potential impact of inadvertent spill of used fryerlator oil. Contact: Land-Tech Consultants, Inc., Playhouse Corner, Suite 205, Southbury, CT 06488 (203-264-8300).
- Reintroduction of saltwater into degraded former salt marshes, removal of dredge material, and restoration of tidal creek in several sites in Connecticut with transect and line intercept monitoring of results. Contact: Long Island Sound Program, Department of Environmental Protection, 165 Capitol Avenue, Hartford, CT 06106 (203-566-7404).
- Annual cutting of perimeter of 1-acre stand and monitoring with aerial photos on 5-year basis; herbicide application on small patch at edge of salt marsh. Contact: The Nature Conservancy, 55 High Street, Middletown, CT 06457 (203-344-0716).

Delaware:

- Aerial spraying of Rodeo™ (glyphosate) and water management plan using stoplogs and vegetation analyses (using transects that measure density and species of plants) of replacement species. Contact: Bombay Hook National Wildlife Refuge, RD #1, Box 147, Smyrna, DE 19977 (302-653-9345).
- Monitoring the ecological factors (water table level, pH, salinity) governing the growth of *Phragmites* in four habitats: open high salt marsh, open low salt marsh, brackish water impoundment, and freshwater impoundment. Investigating *Phragmites* control with glyphosate. Contact: Delaware Division of Fish and Wildlife, P.O. Box 1401, Dover, DE 19903 (302-653-2079).

Louisiana (see Research programs section below)

Massachusetts:

- Cutting three times in one season, followed by opening of tidal floodgate to restore natural water regime, with initial 1-meter random quadrants to measure stem density and plant height. Contact: Department of Public Works, Town of Quincy, Quincy, MA 02169 (617-773-1380 x210) or Ross Dobberteen, Lelito Environmental Consultants, 2 Bourbon St. #102, Peabody, MA 01960 (508-535-7861).
- Aerial spray of Rodeo™ (glyphosate) 2 years in a row, with winter burning; aerial photos to determine decrease in affected boundaries. Contact: Parker National Wildlife Refuge, Northern Blvd. Plum Island, Newburyport, MA 01950 (508-465-5753).
- Clear plastic over cut bands along edge of tidal pond and cutting around perimeter of 0.25-acre stand. Contact: Massachusetts Audubon Society, South Great Road, Lincoln, MA 01773 (617-259-9500).
- Plastic mulch experiments. Contact: Brookline Massachusetts Conservation Commission (617-730-2088).
- Restoration of saltmarshes now dominated by *Phragmites*. Contact: U.S. Army Corps of Engineers, New England Division, 424 Trapelo Road, Waltham, MA 02254 (617-647-8347).

Maryland:

- Nassawango Creek, A Nature Conservancy Preserve, Rodeo™ (glyphosate) applied with backpack sprayer. Monitoring site to determine both reaction of natural plant community and evidence of *Phragmites* re-invasion. Contact: The Nature Conservancy, Chevy Chase Center Office Building, 35 Wisconsin Circle, Suite 304, Chevy Chase, Maryland 20815 (301-656-8073).
- Spraying with Rodeo™ (glyphosate), burning; monitoring vegetation and invertebrates, annual expansion of *Phragmites* in untreated areas. Contact: Environmental Center, Anne Arundel Community College, 101 College Parkway, Arnold, MD 22012-1895 (410-647-7100).

New Jersey:

- Aerial spraying with Rodeo™ (glyphosate), prescribed burn to remove litter, evaluating success. Contact: National Wildlife Refuge, Brigantine Division, P.O. Box 72, Great Creek RD, Oceanville, NJ 08231 (609-652-1665).
- Pulling rhizomes, chemical spray; visual monitoring of presence/absence, sense of height and density. Contact: The Nature Conservancy, 17 Fairmont Road, Pottersville, NJ 07979 (908-439-3007).

New York:

- Cutting (herbicide use would require a permit), using visual assessment for success. Contact: Department of Environmental Conservation, 700 Troy-Schenectady Road, Lathan, NY 12110-2400 (518-783-3932).
- Cutting and covering with plastic (black and clear); dripping herbicide in cut stems with syringe at Constitution Marsh, New York. Contact: Museum of Hudson Highlands, The Boulevard, P.O. Box 181, Cornwall-on-Hudson, NY 12520 (914-534-7781) or National Audubon Society, RFD 2, Route 9D, Garrison, NY 10524 (914-265-2601).
- Open marsh water management with GIS infrared aerial photos and black and white photos (1986 and 1990) to monitor success. Contact: New York DEC, Building 40, SUNY Stony Brook, NY 11790-2356 (516-751-7900 x379 or 516-751-2719)

- Using water level manipulation and burning and visual monitoring. Contact: Wertheim NWR, P.O. Box 21, Smith Road, Shirley, NY 11967 (516-286-0485).

Pennsylvania:

- Tinicum National Environmental Center chemical application, 18 acre restoration with seeding. Contact: Tinicum Environmental Center, Scott Plaza 2, Philadelphia, PA 19113 (215-521-0663).

Ohio:

- Arcola Creek Wetland, Morgan Marsh controlling *Phragmites* by cutting when reserves are in the aerial portion of the plant (before nutrients are translocated into the rhizomes); using aerial photos to map extent of areas, small (1 by 1 meter plots) to measure stem density. Contact: The Nature Conservancy, Ohio Field Office, 1504 West 1st Ave., Columbus, OH 43212 (614-486-6789).

Virginia:

- Rodeo™ (glyphosate) application and monitoring program, with transects (mainly used for changes in vegetation and not in *Phragmites*) and vegetation maps on "topo" scale. Contact: Chincoteague National Wildlife Refuge, Chincoteague, VA 23336 (804-336-6122).
- Winter burns, checking progress in summer with six 400-meter transects perpendicular to the shore that measure percent cover and list species in 0.1 square meter plots every 20 meters; success marginal. Contact: Public Works Office, Building Q29, Aegis Combat System Center, Wallops Island, VA 23337 (804-824-2082).

Research

Management research programs

Louisiana—Aerial photographs of the Mississippi River Delta indicated that different stands of *Phragmites* had different infrared signatures. Isozyme analyses were performed on samples from these stands to determine whether they differed genetically and constituted different clones. Two distinct clones were found, and both differed from stands elsewhere on the gulf coast. Additional isozymal work is planned on populations from elsewhere on the gulf coast and, if time allows, from populations in the Eastern and Great Lakes States as well.

For research on population biology and control methods, refer to Biological monitoring programs section.

Research needs (general)

What are the genetics of natural populations and how do stable and invasive populations differ?

Management research needs

Research on the following facets of *Phragmites* invasions and basic biology are needed:

- What types and levels of disturbance and stress induce *Phragmites* to invade and/or dominate an area?
- How effective are various control programs and what conditions promote or allow *Phragmites* to reinvade areas from which it has been removed?
- If *Phragmites* does reinvade, how long does this process take?
- Are there ways to alleviate or mitigate for the stresses that induce the spread of *Phragmites?*
- Can the use of competitive plantings of *Typha* or other desirable species be used to control *Phragmites*.

References

Ailes, I. 1985. Biologist, Chincoteague National Wildlife Refuge. Telephone conversation with Marianne Marks.

Ailes, I. 1991. Biologist, Chincoteague National Wildlife Refuge. Telephone conversation with Beth Lapin, Nov. 1991.

Ailes, M. 1992. Ecologist, U.S. Navy. Telephone conversation with Beth Lapin, Jan. 1992.

Ailstock, M.S., T.W. Suman, and D.H. Williams. 1990. Environmental impacts, treatment, methodologies and management criteria for establishment of a statewide policy for the control of the marsh plant *Phragmites;* year two. Maryland Dep. Nat. Resourc. Rep., 27 pp. + appen.

Anderson, J. 1992. Biologist, Massachusetts Audubon Society. Telephone conversation with Beth Lapin, Jan. 1992.

Beall, D.L. 1984. Brigantine Division, Marsh vegetation rehabilitation—chemical control of *Phragmites*. U.S. Fish and Wildlife Serv., 8 pp.

Beall, D.L. 1991. Refuge manager, Brigantine National Wildlife Refuge. Telephone conversation with Beth Lapin, Nov. 1991.

Bjork, J. 1967. Ecological investigation of *Phragmites communis*—studies in theoretic and applied limnology. Folia limnologica Scandinavica 14, Lund, Sweden, 248 pp.

Boar, R.R., C.E. Crook, and B. Moss. 1989. Regression of *Phragmites australis* reedswamps and recent changes of water chemistry in the Norfolk Broadland, England. Aquatic Botany 35:41–55.

Bongiorno, S.F., J.R. Trautman, T.J. Steinke, S. Kawa-Raymond, and D. Warner. 1984. A study of restoration in Pine Creek salt marsh, Fairfield, Connecticut. *In* F.J. Webb (ed.), Proc. 11th An. Conf. in Wetlands Restoration and Creation, Hillsborough Community Col., Tampa, Florida.

Boone, J. 1991. University of Georgia, Athens. Telephone conversation with Beth Lapin, Nov. 1991.

Boone, J., E. Furbish, and K. Turner. 1987. Control of *Phragmites communis:* results of burning, cutting, and covering with plastic in a North Carolina marsh. CPSU technical report 41, National Park Service, 15 pp.

Boone, J., E. Furbish, K. Turner, and S. Bratton. 1988. Clear plastic. A non-chemical herbicide. Restoration and Management Notes 6(2):101.

Brandt, S.J. 1983. A health and environmental report on Rodeo™ herbicide. Monsanto Agric. Prod. Co., St. Louis, Missouri, 3 pp.

Breternitz, D.A., C.K. Robinson, and G.T. Gross. 1986. Dolores archaeological program: final synthetic report. U.S. Dep. Interior, Bur. Reclamation, Denver.

Chuzhoza, A.P., and L.Y. Arbuzova. 1970. Specific morphological and biological features of aquatic adventitious roots in *Phragmites communis*.

Clayton, W.D. 1965. The correct name of the common reed. Taxon 17:157–158.

Comes, R.D., V.F. Bruns, and A.D. Kelly. 1976. Residues and persistence of glyphosate in irrigation water. Weed Science 24:47–58.

Cross, D.H., and K.L. Fleming. 1989. Control of *Phragmites* or common reed. U.S. Fish and Wildlife Leaf. 13.4.12, 5 pp.

Daly, P. 1991. Refuge manger, Bombay Hook, Prime Hook National Wildlife Refuges. Telephone conversation with Beth Lapin, Nov. 1991.

Daly, P.D. 1984. Prime Hook Narrative Report. U.S. Fish and Wildlife Serv., 15 pp.

Den, Hartog, C.J. Kvet, and H. Sukopp. 1989. Reed. A common species in decline. Aquatic Botany 35:1–4.

Dobberteen, R. 1991. Wildlife biologist, Lelito Environmental Consultants, Peabody, Maine. Telephone conversation with Beth Lapin, Nov. 1991.

Dowlen, D. 1985. Everglades National Park, Florida. Telephone conversation with Marianne Marks.

Droege, M. 1991. Stewardship director, Maryland office, The Nature Conservancy. Telephone conversation with Beth Lapin, Nov. 1991.

Durska, B. 1970. Changes in the reed (*Phragmites communis Trin.*) condition caused by diseases of fungal and animal origin. Pol. Arch. Hydrobiol. 17:373–396.

Fernald, M.L. 1950. Gray's manual of botany. 8th ed. Corrected printing in 1970 by D. Van Nostrand Company, New York, New York, 1,632 pp.

Folmar, L.C., H.O. Sanders, and A.M. Julin. 1979. Toxicity of the herbicide glyphosate and several of its formulations to fish and aquatic invertebrates. Arch. Environ. Contam. Toxicol. 8:269–278.

Graneli, W. 1989. Influence of standing litter on shoot production in reed, *Phragmites australis* (Cav.) Trin. Ex Steudel. Aquat. Bot. 35:99–109.

Hara, T., J. van der Toorn, and J.H. Mook. 1993. Growth dynamics and size structure of shoots of *Phragmites australis*, a clonal plant. J. Ecol. 81:47–60.

Haslam, S.M. 1968. The biology of reed (*Phragmites communis*) in relation to its control. Proc. 9th BR. Weed Control Cong., pp. 382–387.

Haslam, S.M. 1970. The performance of *Phragmites communis Trin.* in relation to water supply. Ann. Bot. N.S. 34:867–877.

Haslam, S.M. 1971a. Community regulation in *Phragmites communis Trin.* I. monodominant stands. J. Ecol. 59:65–73.

Haslam, S.M. 1971b. The development and establishment of young plants of *Phragmites communis Trin.* Ann. Bot. N.S. 35:1,059–1,072.

Haslam, S.M. 1972. *Phragmites communis Trin.* biological flora British Isles. J. Ecol. 60:585–610.

Hauber, D.P., D.A. White, S.P. Powers, and F.R. DeFrancesch. 1991. Isozyme variation and correspondence with unusual infrared reflectance patterns in *Phragmites australis* (Poaceae). Plant Systematics and Evolution 178:1–8.

Hauber, Dan. 1992. Faculty member, Department of Biological Sciences, Loyola University, New Orleans, Louisiana. Telephone conversation with John Randall, Sep. 1992.

Hayden, A. 1947. Notes on destructive factors operating among the emergent plants of the Ruthven area in the summer of 1947. Qrt. Rep. Iowa Coop. Wildl. Res. Unit. 44:331–343.

Healey, Joann. 1992. Biologist, Parker National Wildlife Refuge. Telephone conversation with Beth Lapin, Jan. 1992.

Hellings, S.E. and J.L. Gallagher. 1992. The effects of salinity and flooding on *Phragmites australis.* J. Applied Ecol. 29:41–49.

Hitchcock, A.S., and A. Chase. 1950. Manual of the grasses of the United States, 2nd ed. USDA Misc. Pub. No. 200. United States Government Printing Office, Washington, DC, 1,051 pp.

Hocking, P.J., C.M. Finlayson, and A.J. Chick. 1983. The biology of Australian weeds. 12. *Phragmites australis* (Cav.) Trin. ex Steud. J. Australian Inst. Agric. Sci., pp. 123–132.

Howard, R., D.G. Rhodes, J.W. Simmers. 1978. a review of the biology and potential control techniques for *Phragmites australis.* U.S. Army Eng. Waterway Exp. Sta., Vicksburg, Mississippi, 80 pp.

Huffman, Mary. 1992. Project director, Lake Wales Project, Florida, The Nature Conservancy. Telephone conversation with Beth Lapin, Jan. 1992.

Johnson, E. 1991. Stewardship director, New Jersey Field Office, The Nature Conservancy. Telephone conversation with Beth Lapin, Nov. 1991.

Johnstone, Rick. 1991. Delmarva Power. Telephone conversation with Beth Lapin, Nov. 1991.

Jones, W.L., and W.C. Lehman. 1987. *Phragmites* control and revegetation following aerial applications of glyphosate in Delaware. *In* W.R. Whitman and W.H. Meredith (eds.), Waterfowl and Wetlands Symp., Delaware Dep. Nat. Resourc. and Environmental Control, Dover, Delaware.

Jontos, Robert Jr. 1992. Consultant, Land-Tech Consultants. Telephone conversation with Beth Lapin, Jan. 1992.

Jontos, Robert Jr., and Christopher P. Allan. 1984. Test salt to control *Phragmites* in salt marsh restoration (Connecticut). Rest. and Mgmt. Notes 2(1):32.

Kartesz, J.T., and R. Kartesz. 1980. A synonymized checklist of the vascular flora of the U.S., Canada and Greenland. Vol. 2, The biota of North America, Univ. of North Carolina Press, Chapel Hill, 500 pp.

Keene, Chuck. 1991. Biologist, Museum of Hudson Highlands. Telephone conversation with Beth Lapin, Nov. 1991.

Kim, K.S., Y.S. Moon, and C.K. Lim. 1985. Effect of NaCl on germination of *Atriplex gmelini* and *Phragmites communis* (in Korean with English abstract). Korean J. Botany 28:253–259.

Klockner, W. 1985. Stewardship director, Maryland chapter, The Nature Conservancy, Telephone conversation with Marianne Marks, June 1985.

Lefor, M.W. 1992. Wetland biologist, University of Connecticut. Telephone conversation and in-person interview with Beth Lapin, Feb. 1992.

Lefor, M.W. 1993. Wetland biologist, University of Connecticut. Letter to John M. Randall, Mar. 1993.

Lehman, W.C. 1984. Project Benchmark. Ecological factors governing growth of *Phragmites* and preliminary investigation of *Phragmites* control with glyphosate. Delaware Div. Fish and Wildlife, 30 pp.

Lehman, W.C. 1992. Biologist, Delaware Division of Fish and Wildlife. Telephone conversation with Beth Lapin, Jan. 1992.

Lynch, J.J., T. O'Neill, and D.E. Lay. 1947. Management and significance of damage by geese and muskrats to gulf coast marshes. J. Wildlife Mgmt. 11:50–76.

Mason, H.L. 1969. A flora of the marshes of California. Univ. Calif. Press, Berkeley, 878 pp.

Matoh, T., N. Matsushita, and E. Takahashi. 1988. Salt tolerance of the reed plant *Phragmites communis.* Physiologia Planarum 72:8–14.

McKee, K.L., I.A. Mendelssohn, and D.M. Burdick. 1989. Effect of long-term flooding on root metabolic response in five freshwater marsh plant species. Can. J. Bot. 67:3446–3452.

McNabb, C.D., and T.R. Batterson. 1991. Occurrence of the common reed, *Phragmites australis*, along roadsides in lower Michigan. Michigan Academician 23:211–220.

Metzler, K., and R. Rozsa. 1987. Additional notes on the tidal wetlands of the Connecticut River. CT Bot. Soc. Newsletter 15:1–6.

Monsanto Company. 1985. Rodeo aquatic herbicide; complete directions for use in aquatic sites. St. Louis, Missouri, 3 pp.

Mook, J.H., and J. van der Toorn. 1982. The influence of environmental factors and management on stands of *Phragmites australis.* II. Effects on yield and its relationships with shoot density. J. Appl. Ecol. 19:501–517.

Niering, W.A., and R.S. Warren. 1977. Our dynamic tidal marshes: vegetation changes as revealed by peat analysis. Connecticut Arboretum Bul. 12, 22 pp.

Niniviaggi, Dominick. 1991. Marine resources specialist, Bureau of Marine Habitat Protection, Department of Environmental Conservation, Stoney Brook, New York. Telephone conversation with Beth Lapin, Nov. 1991.

Nugent, R. 1991. Director, Tinicum Environmental Center in John Hindes National Wildlife Refuge, Pennsylvania. Telephone conversation with Beth Lapin, Nov. 1991.

Ostendorp, W. 1989. 'Die-back' of reeds in Europe—a critical review of literature. Aquatic Botany 35:5–26.

Ostendorp, W. 1991. Damage by episodic flooding to *Phragmites* reeds in a prealpine lake: proposal of a model. Oecologia 86:119–124.

Osterbrock, A.J. 1984. *Phragmites australis.* The problem and potential solutions. Ohio Field Office, Stewardship, 8 pp.

Parris, R. 1991. Biologist, Wertheim National Wildlife Refuge, Shirley, New York. Telephone conversation with Beth Lapin, Nov. 1991.

Penko, J.M. 1985. Ecological studies of *Typha* in Minnesota: Typha-insect interactions, and the productivity of floating stands. M.S. thesis, Univ. Minnesota.

Penko, J.M. 1993. Ecologist, U.S. Army Corps of Engineers, Waltham, Massachusetts. Letter to John M. Randall, Apr. 1993.

Pintera, A. 1971. Some observations on mealy plum aphid, Hyalopterus pruni Geoffi, occurring on reeds. Hidrobiologia (Bucharest) 12:293–295.

Pokorny, B. 1971. Flies of the genus Lipara meigen on common reed. Hidrobiologia (Bucharest) 12:287–292.

Poole, J. 1985. Botanist, Texas Natural Heritage Program, The Nature Conservancy. Letter to Marianne Marks, Mar. 1985.

Raicu, P., S. Staicu, V. Stoian, and T. Roman. 1972. *Phragmites communis Trin.* complement in the Danube Delta. Hydrobiologia 39:83–89.

Rawinski, T. 1985. Common reed (*Phragmites australis*) in a select group of New York/New England natural areas, an overview. Eastern Heritage Task Force, The Nature Conservancy, 6 pp.

Rawinski, T. 1985. Ecologist, Eastern Heritage Task Force, The Nature Conservancy. Conversation with Marianne Marks, July 1985.

Rawinski, T. 1992. Ecologist, Virginia Heritage Program. Telephone conversation with Beth Lapin, Feb. 1992.

Reimer, D.N. 1973. Effects of rate, spray volume, and surfactant on the control of *Phragmites* with glyphosate. Proc. N.E. Weed Sci. Soc. 27:101–104.

Ricciuti, E.R. 1983. The all too common, common reed. Audubon Magazine, Sep. 1983, pp. 65–66.

Rod, J. 1992. Manager, Constitution Marsh, National Audubon Society. Telephone conversation with Beth Lapin, Feb. 1992.

Rodewald-Rudescu, L. 1974. Das Schilfrohr. Die Binnengewasser, No. 27. E. Scweiterbartsche Verlagbuchhanlung, Stuttgart, Germany, 294 pp.

Roman, C.T., W.A. Niering, and R.S. Warren. 1984. Salt marsh vegetation change in response to tidal restriction. Environ. Man. 8:141–150.

Rozsa, R. 1992. Biologist, Department of Environmental Protection. Telephone conversation with Beth Lapin.

Schneider, K. 1991. Coordinator, New York Heritage Program. Telephone conversation with Beth Lapin.

Seidel, T. 1992. Stewardship and volunteer coordinator, Ohio office, The Nature Conservancy. Telephone conversation with Beth Lapin.

Skuhravy, V. 1978. Invertebrates: destroyers of the common reed. *In* D. Dykyjova and J. Kvet (eds.), Pond Littoral Ecosystems Structure and Functioning. Springer-Verlag, pp. 376–388.

Sorrie, B. 1985. Botanist, Massachusetts Natural Heritage Program. Letter to Marianne Marks, Apr. 1985.

Stark, H., and M. Dienst. 1989. Dynamics of lakeside reed belts at Lake Constance (Untersee) from 1984 to 1987. Aquatic Botany 35:63–70.

Steinke, T. 1992. Conversation commissioner, Fairfield, Connecticut. Telephone conversation with Beth Lapin, Aug. 1992.

Sukopp, H., and B. Markstein. 1989, Changes of the reed beds along the Berlin Havel, 1962–1987. Aquatic Botany 35:27–39.

Thompson, D.J., and J.M. Shay. 1985. The effects of fire on *Phragmites australis* in the Delta Marsh, Manitoba. Can. J. Bot. 63:1964–1869.

Thompson, D.J., and J.M. Shay. 1989. First-year response of a *Phragmites* marsh community to seasonal burning. Can. J. Bot. 67:1448–1455.

Truitt, Barry. 1992. Steward, Virginia Coast Reserve, Nassawadox, Virginia. Interview with John Randall, Dec. 1992.

Tscharntke, T. 1988. Variability of the grass *Phragmites australis* in relation to the behavior and mortality of the gall-inducing midge Giraudiella inclusa (*Diptera*, *Cecidomyiidae*). Oecologia 76:504–512.

Tucker, G.C. 1990. The genera of Arundinoideae (*Gramineae*) in the Southeastern United States. J. Arnold Arboretum 71:145–177.

Turner, K. 1992. Chief of resource management, Lake Mead Recreation Area, Nevada. Telephone conversation with Beth Lapin, January 1992.

van der Merff, M., J.W. Simmers, and S.H. Kay. 1987. Biology, management and utilization of common reed *Phragmites australis.* U.S. Army rep., contract no. DAJA45-86-M-0482, 101 pp.

van der Toorn, J., and J.H. Mook. 1982. The influence of environmental factors and management on stands of *Phragmites australis.* I. Effects of burning, frost and insect damage on shoot density and shoot size. J. Appl. Ecol. 19:477–499.

van Deursen, E.J.M., and H.J. Drost. 1990. Defoliation and treading by cattle of reed *Phragmites australis.* J. Appl. Ecol. 27:284–297.

Weisner, W.E.B., and W. Graneli. 1989. Influence of substrate conditions on the growth of *Phragmites australis* after a reduction in oxygen transport to belowground parts. Aquatic Bot. 35:71–80.

Wheelwright, M. 1991. Department of Public Works. Quincy, Massachusetts. Telephone conversation with Beth Lapin, Nov. 1991.

Young, J. 1985. Land Steward Florida Field Office, The Nature Conservancy. Telephone conversation with Marianne Marks, May 1985.

III.J.2.f Control and management of North American cattail species (*Typha* spp.)

(J. Bender, The Nature Conservancy, Arlington, Virginia, author; Jay Rendall, The Nature Conservancy, Arlington, Virginia, update January 11, 1987, and revised 1998; K. Motivans, document preparation and maintenance, S. Apfelbaum Applied Ecological Services, Inc., Juda, WI 53550 608/897-8547; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001. Distribution map and photographs, NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

This abstract provides information on the biology and control of invasive plant species in wetlands.

To the user

Element Stewardship Abstracts (ESAs) are prepared to provide The Nature Conservancy's Stewardship staff and other land managers with management-related information on those species and communities that are most important to protect, or most important to control. The abstracts organize and summarize data from numerous sources including literature and researchers and managers actively working with the species or community.

We hope, by providing this abstract free of charge, to encourage users to contribute their information to the abstract. This sharing of information will benefit all land managers by ensuring the availability of an abstract that contains up-to-date information on management techniques and knowledgeable contacts. Contributors of information will be acknowledged within the abstract and receive updated editions. To contribute information, contact the editor whose address is listed above.

For ease of update and retrievability, the abstracts are stored on computer at the national office of The Nature Conservancy. This abstract is a compilation of available information and is not an endorsement of particular practices or products.

Please do not remove this cover statement from the attached abstract.

1815 North Lynn Street, Arlington, Virginia 22209 (703) 841 5300

Contents

The Nature Conservancy
Element Stewardship Abstract
North American Cattail species (*Typha* spp.)

Identifiers

Scientific name: *Typha* spp.
Common name: Cattail

Description

The cattail genus (*Typha* spp.) is an erect, perennial freshwater aquatic herb that can grow 3 or more meters in height. The linear cattail leaves are thick, ribbon-like structures that have a spongy cross section exhibiting air channels. The subterranean stem arises from thick, creeping rhizomes. North American cattails have minute, brown, male flowers (staminate) thickly clustered on a club-like spadix. The lower part of the spadix bares the female flowers (pistillate). Three species and several hybrids in the cattail genus occur in North America (Smith 1961, 1962, 1967). *Typha latifolia*, broadleaf cattail, is distinguished from *T. angustifolia*, narrowleaf cattail, by the relative width of the leaf and the position of the staminate and pistillate portions of the spadix (heads).

Typha latifolia has 6- to 23-millimeter-wide leaves that are flat, sheathing, and pale grayish-green. T. angustifolia has 3- to 8-millimeter-wide leaves that are full green and somewhat convex on back (ARS 1971). In *T. latifolia* the staminate and pistillate heads are contiguous or nearly so, whereas in *T. angustifolia* the heads are separated by about 3 centimeters. Cattail fruits differ among the two major species. *T. angustifolia* fruits are about 5 to 8 millimeters long with hairs arising above the middle. *T. latifolia* fruits are about 1 centimeter long with hairs arising near the base (ARS 1971). The tall cattail (*Typha domingensis*) may be difficult to separate from *T. angustifolia*. *T. domingensis* is usually taller and has flattened and more numerous leaves (Apfelbaum 1985). Hybrids of intermediate appearance have been reported and are often referred to as the species *Typha* x *glauca*.

Typha latifolia

Elements included in EMG
Typha latifolia (Broadleaf cattail)
Typha angustifolia (Narrowleaf cattail)
Typha x *glauca* (Hybrid cattail)
Typha domingensis (Southern cattail)

Stewardship summary

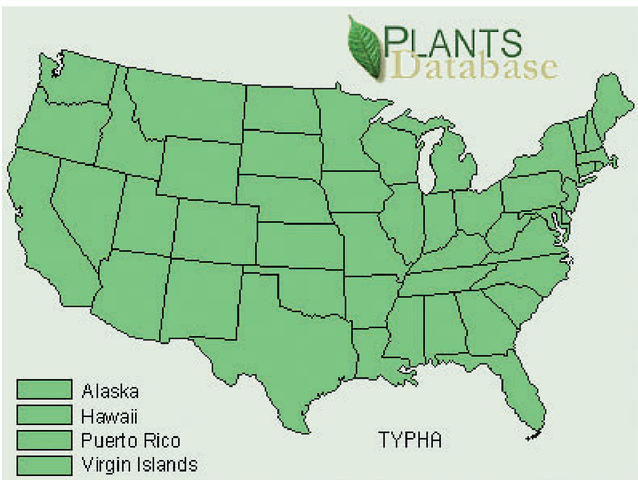
Cattail control is an important consideration for natural areas. Monitoring the spread of cattails by aerial surveys and sampling transects can help determine the extent of cattail monocultures. Research into new biological control methods and the recovery of communities after cattail management needs to be conducted. Control techniques of fire and physical removal (cutting) in conjunction with flooding are most appropriate.

Natural history

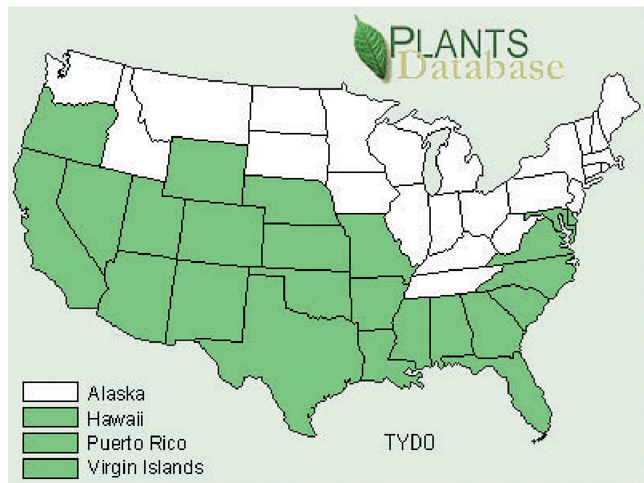
Range

Cattails have a cosmopolitan distribution and wide ecological amplitude.

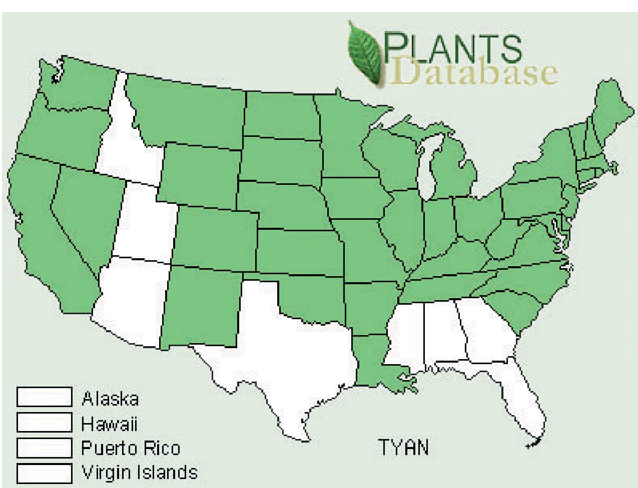
Typha latifolia (broadleaf cattail) is found throughout North America from sea level to 2134-M (7,000 ft) elevation.



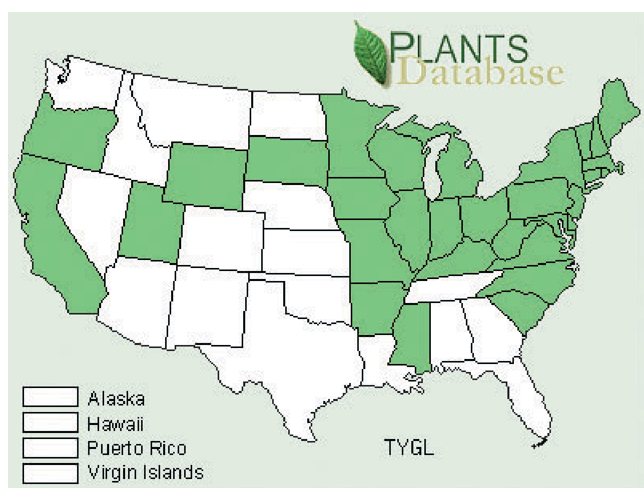
Typha domingensis (southern cattail) has a range that extends from the Southwest United States, southern California, and east to southeastern Virginia.



Typha angustifolia (narrowleaf cattail) is widely distributed in the Eastern and Northern United States.



Typha x *glauca* (a hybrid cross) is widely distributed in the East as well as extending into the Northern Plains and Western United States.



Habitat

Typha sp. can be found in wetlands, sedge meadows, along slow moving streams, riverbanks, and lake shores. The plant is in areas of widely fluctuating water levels, such as roadside ditches, reservoirs, and other disturbed wet soil areas. Cattails commonly invade the pelagic zones of bogs (Gustafson 1976). Typical associates include *Phragmites australis*, *Lythrum salicaria*, *Spartina* sp., *Acorus calamus*, *Scirpus* sp., and *Sagittaria latifolia*. *Typha angustifolia* is generally restricted to unstable environments, often with basic, calcareous, or somewhat salty soil (Fassett and Calhoun 1952). Narrow-leaved cattail can grow in deeper water compared to *T. latifolia*, although both species reach maximum growth at a water depth of 20 inches (50 cm) (Grace and Wetzel 1981). A robust hybrid between narrowleaf and broadleaf cattail, *Typha* x *glauca*, has similar habitat requirements to *T. angustifolia*.

Typha latifolia is the only species of cattail usually found in relatively undisturbed habitats throughout North America (Smith 1967). The tolerance of *T. angustifolia* to high concentrations of lead, zinc, copper, and nickel has been demonstrated (Taylor and Crowder 1984). This species has been employed in secondary wastewater treatment schemes (Gopal and Sharma 1980).

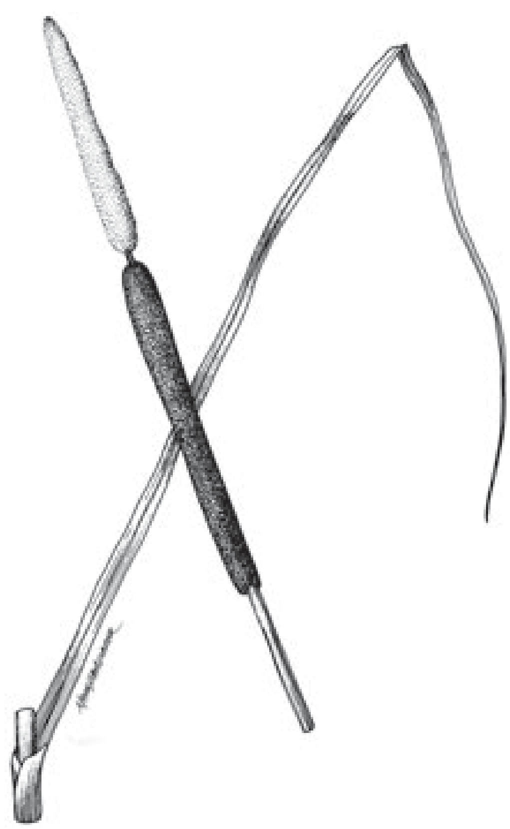
Typha latifolia is found in the most favorable sites where it competes against other species. *T. angustifolia* and *T. domingensis* are restricted to less favorable and more saline habitats where they occur with *T. latifolia* (Gustafson 1976). *Typha latifolia* often displaces *T. angustifolia* in shallow (up to 6 in) water, restricting the latter species to deep water (Grace and Wetzel 1981). *Typha angustifolia* is considered a pioneer in secondary succession of disturbed bogs (Wilcox et al. 1984). Presumably, an increase in the acidity of a bog would lower the pH and reduce the invasion of *T. angustifolia.* Theodore Cochrane (pers. comm.) of the University of Wisconsin-Madison herbarium states that most early herbarium specimens are *T. latifolia* and only recently have *T. angustifolia* specimens been collected from Wisconsin wetlands.

Cattails can grow on a wide gradient of substrate types. Wet pure sand, peat, clay, and loamy soils have been documented under cattail stands. Worldwide distribution of cattails is summarized by Morton (1975).

Ecology

Cattails flower in late May and June and sometimes later (up to late July) depending, perhaps, on soil and water temperatures as influenced by climate and litter in a stand. The wind-borne pollen attaches to stigmas of female florets to eventually produce achene fruits. The elongated embryo and stalk are covered with fine, unmated hairs that aid in wind dispersal. Fruits are mature in August and September. Seeds are very small, weighing 0.055 milligram each (Keddy and Ellis 1985).

Many cattail germination studies have been conducted. Some suggest that germination requirements are few. Seed germination can be 100 percent in slightly flooded conditions (Smith 1967). *Typha latifolia* seeds are less tolerant to salt (NaCl) concentrations in the substrate when compared to *T. angustifolia* seeds. However, seeds of both species that had been soaked in salt solution would germinate after being returned to nonsaline conditions (McMillan 1959). *Typha angustifolia* seeds showed no significant germination response when sprouted along a moisture



Narrowleaf cattail

gradient that ranged from 2 inches below substrate to 4 inches above (Keddy and Ellis 1985). Other studies have confirmed that water is required at a depth of 1 inch for germination. Sifton (1959) showed light and low oxygen tensions affected germination of broad-leaved cattail.

Van der Valk and Davis (1976) suggested that the germination of *Typha* seeds could be inhibited by an allelopathic interaction caused by *Typha* litter. Seed longevity and dormancy may be affected by soil moisture, temperature, and soil atmosphere (Schafer and Chilcote 1970, Roberts 1972, Meyer and Poljakoff-Mayber 1963, Morinaga 1926).

Young *Typha* shoots grow rapidly from seeds in favorable substrates. Cattail colonies are commonly maintained by vegetative reproduction. A perennial rootstock is the major organ responsible for reproduction (Apfelbaum 1985). Cattail productivity has been well documented. Net annual production is generally estimated as the maximum standing crop (shoot biomass) values for a good site of between 10 to 18 ounces per foot (dry weight) (Gustafson 1976). Figures for *Typha* production mostly exceed the average standing crop yields for maize and sorghum.

Shoot density reports (numbers of stems per square feet) range from 8.5 per square feet (Curtis 1959) in Wisconsin to an extreme example reported by Dykyjova, et al. (1971) of 33 per square feet. In a greenhouse experiment, 98 vegetative shoots and 104 crown buds were produced on a single seedling during its first year (Timmons et al. 1963). Cattails can produce 20,000 to 700,000 fruits per inflorescence (Prunster 1941, Marsh 1962, Yeo 1964). Vegetative growth by broad-leaved cattails of 17 feet (518 cm) annually have been recorded (McDonald 1951), and plants grown from seed flowered the second year (Smith 1967, Yeo 1964).

Cattail plants produce a dense rhizome mat, and the clustered leaves produce a thick litter layer. Dense cattail growth and litter may reduce the opportunity for other plants to establish or survive (Wesson and Waring 1969).

Calculations show that a natural stand of cattails may fix 16 pounds nitrogen per acre per year, or about 8 percent of the total nitrogen present in the standing crop (Biesboer 1984).

The structure of cattail stands as it is, with upright leaves, high leaf area, balanced horizontal and vertical distribution of leaf area, and shifts in leaf angle, are all factors that permit monoculture success. An open, generously sunny habitat and abundant moisture can provide the setting for maximum cattail production.

Typha plants are mined by caterpillars of the moths *Arzama obliqua* and *Nonagria oblonga* (Klots 1966). Aphids and *Colandra pertinaux* (the snout beetle) also feed on *Typha* leaves and stems. The stems may have many species of pupa living within them (Klots 1966). The cattail rhizomes provide food to mammals, such as the muskrat. The grazing of muskrats may greatly influence cattail communities. A cycling population of muskrats may reach such a density so as to totally set back a cattail stand for the season. These “eat outs” are important to maintain open water in a balanced system. Muskrats use leaves and stems for houses and eat the rhizomes (Zimmerman, pers. comm. 1987). Cattail fruits provide nesting material for terrestrial birds, and dry stems may be used by aquatic birds. Aboveground portions die in the late fall and rhizomes over winter. In Wisconsin, it was found that average winter marsh temperatures of more than 8 degrees Celsius reduced carbohydrate reserves in *Typha latifolia* to an extent sufficient to inhibit shoot growth in the spring (Adriano et al. 1980). Cattail population success has been correlated with nutrient fertility, water level, and substrate temperature (Adriano et al. 1980).

The plant tissues can store relatively high concentrations of some metals. *Typha* appears to have an internal copper and nickel tolerance mechanism. It is not likely that there is an evolutionary selection for heavy metal tolerance, but rather it is inherent in the species (Taylor and Crowder 1984).

Impacts

Cattail management may be desired in situations where cattails have responded to wetland disturbance by growing in dense monocultures. The genus *Typha* can behave like aggressive introduced weeds in a variety of natural communities throughout North America (Apfelbaum 1985). Cattails are considered serious weeds in some countries (Holm et al. 1979, Morton 1975), but not necessarily in North America. In high-quality natural communities, cattails usually occur as scattered sterile plants (Apfelbaum 1985).

With disruptions to a community, cattail populations may respond by spreading vegetatively at a rapid rate. The effect of the growth spurt is closing open water, eliminating habitat and species diversity, and reducing the opportunity for other plants to become established and survive. Shading is a significant effect on other plants. Cattails are successful because they form extensive monocultures rapidly through vegetative reproduction and maintain their dominance with the formation of dense rhizomes mats and litter.

Cattails have a wide ecological amplitude compared to other species (Pianka 1973). They are tolerant to habitat changes, pollutants in the water system, and saline or basic substrates. A study in Indiana concluded that the three basic events precede the growth of cattails monocultures:

- modified surface hydrology,
- wildfire suppression, and
- wetland enrichment (Wilcox et al. 1984).

Claims that hybrid cattails are responsible for monoculture growths have not been confirmed.

Management/monitoring

Management requirements

Cattails are often purposefully encouraged in some areas to stabilize shorelines from wave action erosion or ice heaving. Two-thirds of wave energy dissipates in 2 meters of cattail beds (Bonham 1983). Cattails have been used to reduce salinity in rice fields (Marsh 1962) and have been considered “scrubbers” in polluted aquatic systems (Gopal and Sharma 1980). Commercial uses of cattails include footwear, roofing, and floor mats. The species has been considered as an important source of protein (Morton 1975) and a fuel source. The objective of management is not to eradicate cattails, but rather to control their spread in natural communities. Specifically, the goals of management should be to:

- Control the spread and domination of potential habitat by cattail in and adjacent to natural areas.
- Circumvent declines in other plant species with cattail proliferation.
- Prevent development of monotypic cattail growth and loss of habitat heterogeneity (Martin et al. 1957).

Management of cattails should be site specific and could include such active measures as handcutting rootstalks, burning and flooding, or shading.

Water level modification

High water conditions in a cattail stand can affect the growth of seedlings, break off mature stalks, or be followed by the immigration of muskrats, which eat the cattail (Zimmerman pers. comm. 1987). The effect of flooding on cattails is not always negative; plants have been known to float up and continue growing until water returns to previous lower levels.

As with any control measure, temporary conditions, such as flooding, do not prevent later seed establishment. Cattail seeds can arrive from a great distance, and it does not take but a few seeds to germinate and rapidly produce clones as adults. The cost of management actions should be considered when dealing with unknown response variables.

Low water conditions, maintained by draining a wetland, significantly affects the overall community (Mallik and Wein 1985). Harris and Marshall (1963) concluded that draining techniques have possible detrimental effects because the plant composition of a wetland can be radically changed. Draining alone can cause a significant increase in *Typha* cover under some conditions (Mallik and Wein 1985). However, to inhibit *Typha* growth, a wetland can be drained and then burned during the summer. If there is no reserve of water over winter, cattails do not survive the following spring, according to Zimmerman (pers. comm. 1987), but there have been no controlled experiments to show this.

Two years of 65-centimeter-deep (26 in) flooding was required before established cattail began to die and open water conditions were created at Sinnissippi Marsh. Cattail initially survived flooding from 1973 to 1977 and became the dominant emergent plant. A light green color, noticeably narrower leaves, and absence of fruiting heads indicated stress in 1976. Cattail stem densities declined 57 percent with all emergent plants dead in 1977. Horicon Marsh, flooded to a depth of 40 centimeters (16 in), showed declines in emergent and aquatic plants. Cattail required 2 years before it declined (Wisconsin DNR 1969 and 1971).

Mature *T. latifolia* and seedlings less than 1 year old are killed by water depths of 25 inches (63.5 cm) and

18 inches (45 cm) or more, respectively. Narrowleaf cattail was unaffected by this degree of flooding. Narrowleaf cattail establishment was prevented when water levels were maintained at 47 inches (1.2 m) or deeper (Steenis et al. 1958). Dryer conditions allowed more clones of *T. angustifolia* to be spread (McMillan 1959).

Because cattails can transpire significant quantities of water (2 to 3 quarts of water/ac/yr) (Fletcher and Elmendor 1955, Zohary 1962), their establishment may serve to exacerbate water level instability and further contribute to disruptive influences supporting increased cattail. Flooding must account for evapotranspiration losses of water to maintain a level effective in cattail control.

Chemical control

For designated preserves or natural areas, especially where system-orientated stewardship is used, chemical applications may not be appropriate. This is particularly true because cattail is an element of certain natural communities. However, use of chemicals to control an overabundance of cattail may have certain applications. Spraying Dalpan (Nelson and Dietz 1966) at 4 to 16 pounds per acre (8.8–35.3 kg/ac) produced 74 to 97 percent reductions in cattails 10 months after a mowed area was sprayed. Cattail regrowth was sprayed at a 24- to 36-inch (58- to 90-cm) height in September. Control was most effective when treated areas could be flooded to 4 to 5 inches (10–15 cm) or deeper. Dalpan spray achieved varied success, but greatest control occurred where cattail stems were cut below water depth regardless of the herbicide quantity used. Poorest results were attained in areas that have shallow fluctuating water levels. Spraying mature cattails rather than regrowth after cutting gave better results. Weller (1975) had similar results with spraying where Amitrol, Rodopan, and Doupon herbicides were effective in creating and maintaining openings for at least 3 years after spraying, but areas were quickly invaded by peripheral cattail. High doses of MCPA or 2,4-D in diesel oil (2.2–4.5 kg/ac) were effective if applied during flowering. Dalpan (9 kg/ac) and Aminotriazole (0.91–1.36 kg/ac) gave good control results in Montana (Timmons et al. 1963). Herbicide applications were found necessary for up to 3 years in some areas. Similar results were found by Grigsby et al. (1955), Heath and Lewis (1957), Krolikowska (1976), Pahuja et al. (1980), Singh and Moolani (1973), and Wisconsin Department of Natural Resources (1969).

Wick and spray applications of Roundup™ followed by manual clipping of all cattail stems was the treatment conducted by Applied Ecological Services and All Services Company (1985) at a pond in northern Illinois. Cattail seeds were just at ripening stage at the time of treatment. Retreatment of Roundup™ several weeks later and subsequent die-off proved this method successful.

Herbicide treatment at flowering may stress the cattail plants more than at other stages since the energy investment by the plant has been channeled into flowering.

Physical control

Hand or mechanical cutting of cattails followed by submergence of all cattail stems results in high control. Up to 100 percent cattail control was measured two growing seasons after treatment. No visible cattail regrowth occurred in 1 year, and cattail rhizomes were dead. The highest cattail control of any method tested was achieved by two clippings followed by stem submergence to at least 3 inches (7.5 cm) (Nelson and Dietz 1966). Control was best if plants were cut in late summer or early fall.

In Iowa (Weller 1975), cutting cattail and reflooding with at least 3.1 inches (8 cm) of standing water over plant stems was effective. Weller also found clipping cattails too early in the growing season (e.g., May) stimulated their growth and resulted in a 25 percent increase in stem counts the following year, with an eventual decline to pre-clip levels. August clipping controlled up to 80 percent of cattail only if followed by submergence. To achieve this control, all cattail stems must be removed. Cutting shoots below the water surface two or three times in one growing season before flower production reduced a cattail stand by 95 to 99 percent in Montana and Utah (Stodola 1967). Similar results were demonstrated by Shekhov (1974) and Sale and Wetzel (1983).

When shoots are cut below the water level, nearly all the oxygen is consumed in a short time, necessitating anaerobic respiration. In *Typha*, ethanol is produced accompanied by tissue breakdown after an oxygen shortage. *Typha* is ill adapted to deprivation of oxygen. Cuttings later than flowering stage are effective only in preventing regrowth for that year and may have no effect on subsequent years (Shekhov 1974).

Cattail control by injuring developing rhizomes and shoots was investigated (Weller 1975). Crushing and reflooding showed that cattails injured after June had poor recoveries. Success of crushing depended on the load used, number of times an area was crushed, and standing water depths after treatment. Spring and early summer treatments generally created favorable seedbeds for cattail and required a fall crushing to control seedlings. Crushing involved pulling a 55-gallon water-filled drum behind a tractor. Deeper water areas showed highest control (up to 100 percent) while regrowth occurred in shallow areas. Although not practical for natural areas management, disking (Weller 1975) and blasting (Nelson and Dietz 1966) have also been investigated as methods of cattail control.

Prescribed burning

Fire alone was found to provide little or no cattail control (Nelson and Dietz 1966). Fires that destroyed cattail roots offered control; however, most fires only burned aboveground biomass and did little to control cattail. Drying in readiness for burning was effective cattail control when done for 2 years in arid Utah. Water was pumped from wetlands, and then cattail stands were allowed to sun dry.

Water level drawdown, burning (spring, fall, and mid-growing season), and reflooding to 8- to 18-inch (20- to 35-cm) water depth or deeper controlled cattail. Fire was found useful for cattail litter cleanup and assisted access for mowing or handclipping (Nelson and Dietz 1966, Weller 1975, Mallik and Wein 1985). **Before providing assistance with a burn or recommending a burn, consult the NRCS burn policy in the General Manual.**

Shading

Black polyethylene tarps were used to cover cattails in an attempted control measure (Nelson and Dietz 1966). Actively growing cattail tips were killed when completely covered for at least 60 days. Greatest control was achieved in July when food resources of cattail were presumed to be lowest (Linde et al. 1976). Problems with holding tarps down and their degradation confounded this investigation. Cattail is generally not shade tolerant.

Monitoring

Cattail control or reduction may be desirable where noticeable increases threaten natural plant diversity and habitat heterogeneity. Increases in the rate of spread and growth of a colony may signal management action. The establishment of cattails in nonwetland areas should be monitored. Gross area monitoring is necessary to determine the effects of management practices and the need for future management.

Aerial surveys are used to document by photographs the spread of cattail colonies (Wilcox et al. 1984). The advance of cattail clones can also be documented by placing permanent markers at the leading edge of colonies. Sampling along shore to water transects using 1 square meter quadrants allows an estimate of percent cover, stem density, and importance value of species. Shore to water transects with the line intercept methods show changes in density and spread.

Monitoring programs:

Contact: Cowles Bog Wetland Complex, Indiana Dunes National Lakeshore, Porter, Indiana 46304.

Contact: University Bay Marsh, Institute of Environmental Studies, University of Wisconsin-Madison, Madison, Wisconsin 53706.

Contact: Pinhook Bog, Indiana.

Contact: Horicon Marsh, Horicon, Wisconsin. Local Department of Natural Resources managers.

Contact: Chicago Botanical Garden, Glencoe, Illinois (restoration work on wetlands).

Contact: Biology Department, Cornell, Ithaca, New York.

Contact: Applied Ecological Services, Inc., N673 Mill Road, Juda, Wisconsin 53550 (monitoring in several dozen wetlands).

Contact: Indiana Field Office, The Nature Conservancy.

Management research needs

Research objectives in the past have concentrated on the effect of cattails on waterfowl production, sewage treatment, fuel production, or recreational opportunities. Few studies on the methods of control of cattails in designated nature preserves or natural areas have been conducted. More effort needs to be put into research with biological diversity and natural area maintenance as the major objectives.

Biological control has not been documented or researched. The effects of shading, day length, or varying light intensity on cattail reproduction is largely unknown (Apfelbaum 1985). Data are not available to test the concerns that a fire used to control or destroy *Typha* rhizomes would destroy other plants or the wetland seedbank. Recent evidence (Apfelbaum unpub. data) suggests repeated annual spring burning in cattail dominated systems stimulates Cyperaceous seed germination even beneath a dense cattail canopy. Whether this is related to litter removal, actual fire scarification, or other causes is unknown. More case studies and data related to the recovery of the natural community after cattail control, particularly fall burning, are important needs for future study. The interactions between animals, water level, and cattail growth need to be studied (Zimmerman pers. comm. 1987). Cost effectiveness of the various methods available for cattail control is an important consideration.

Bibliography

Addy, C.E., and L.G. MacNamara. 1948. Waterfowl management areas. Wildlife Management Institute, Washington, DC, 80 pp.

Adriano, D.C., A. Fulenwider, R.R. Shariz, T.G. Ciraudlo, and G.D. Hoyt. 1980. Growth and mineral nutrition of cattail (*Typha*) as influenced by thermal alteration. J. Environ. Quality 9(4):649–653.

Ahlgren, I.F., and C.E. Ahlgren. 1960. Ecological effects of forest fire. Bot. Rev. 26:483–533.

Apfelbaum, S.I. 1985. Cattail (*Typha* spp.) management. Natural Areas J. 5(3):9–17.

Apfelbaum, S.I., K. Heiman, J. Prokes, D. Tiller, and J.P. Ludwig. 1983. Ecological condition and management opportunities for the Cowles Bog National Natural Landmark and Great Marsh, Indiana Dune National Lakeshore, Porter, Indiana. Rep. Indiana National Lakeshore.

Applied Ecological Services and All Services Company. 1985. Report on effects to control cattails (*Typha angustifolia* and *T. latifolia*) at the Swain family pond. Libertyville, Illinois. Unpublished report.

Bayly, I.L., and T.A. O'Neill. 1972. Seasonal ionic fluctuations in *Typha glauca* community. Ecol. 53(4):714–719.

Bedford, B.I., E.H. Zimmerman, and J.H. Zimmerman. 1974. The wetlands of Dane County. Wisconsin Dept. Nat. Resour. Res. Rep. No. 30, 62 pp.

Bedish, J.W. 1964. Studies of the germination and growth of cattail in relation to marsh management. Masters thesis, Iowa State Univ., Ames, Iowa.

Bedish, J.W. 1964. Cattail moisture requirements and their significance to marsh management. Am. Midl. Nat. 78:288–300.

Bellrose, F.C., and L.G. Brown. 1941. The effect of fluctuating water levels on the muskrat population of the Illinois River Valley. J. Wildl. Mgt. 5:206–212.

Biesboer, D.D. 1984. Nitrogen fixation associated with natural and cultivated stands of *Typha latifolia* L. (*Typhaceae*). Amer. J. Bot. 71(4):505–511.

Bonasera, J.J., and M.A. Leck. 1978. An allelopathy study of marsh plants and soils. Bul. N.J. Acad. Sci. 23(2):83.

Bonham, A.J. 1983. The management of wave-spending vegetation as bank protection against boatwash. Landscape Planning 10:15–30.

Cochrane, T.D. 1987. Botanist, University of Wisconsin at Madison. Personal communication with S. Apfelbaum, K. Motivans, June 1987.

Curtis, J.T. 1959. The vegetation of Wisconsin. University of Wisconsin Press, Madison, Wisconsin.

Dane, C.W. 1956. The succession of aquatic plants in small artificial marshes in New York State. New York Fish and Game J. 6:57–76.

Dudinskii, Y.A., and V.M. Bazhutina. 1976. Leaf growth aspects of *Typha-latifolia* and *Sparganium-Polyedrum* in the initial stages of their development. Bot. Z. (Leningrad) 61(2):263–266.

Dykyjova, D., K. Veblr, and K. Priban. 1971. Productivity and root/shoot ratio of reed swamp species growing in outdoor hydroponic cultures. Folia Geobot. Phytotax., Praha6 233–254.

Fassett, N.C., and B. Calhoun. 1952. Introgression between *Typha latifolia* and *T. angustifolia.* Evolution 6:267–379.

Finlayson, C.M. 1984. Short-term responses of young *Typha domingensis* and *Typha orientalis* plants to high levels of potassium chloride. Aquatic Bot. 20:75–85.

Fletcher, H.C., and H.B. Elmendorf. 1955. Phreatophytes—A serious problem in the west. U.S. Dep. Agric. Yearbook, pp. 423–429.

Giltz, M.L., and W.D. Myser. 1954. A preliminary report on an experiment to prevent cattail die-off. Ecology 35:418.

Gleason, H.A. 1957. The New Britton and Brown illustrated flora of the Northeastern U.S. and adjacent Canada. New York Botanical Garden, New York.

Gopal, B., and K.P. Sharma. 1980. Aquatic weed control versus utilization. Econ. Bot. 33:340–346.

Grace, J.B., and R.G. Wetzel. 1981. Effects of size and growth rate on vegetative reproduction in *Typha. Oecologia* (Berlin) 50(2):158–161.

Grigsby, B.H., C.A. Reimer, and W.A. Cutler. 1955. Observations on the control of cattail *Typha* spp. by chemical sprays. MI Quar. Bul. 37(3):400–406.

Gustafson, T.D. 1976. Production, photosynthesis and the storage and utilization of reserves in a natural stand of *Typha latifolia* L. Ph.D. thesis, Univ. Wisconsin-Madison, 102 pp.

Harris, S.W., and W.H. Marshall. 1963. Ecology of water-level manipulations on a northern marsh. Ecology 44:331–343.

Heath, R.G., and C.R. Lewis. 1957. Aerial control of cattail with radapon. The Dow Chemical Co., Midland, MI, Down to Earth.

Heywood, V.H. 1978. Flowering plants of the world. Mayflower Books, New York, New York, 335 pp.

Hogg, E.H., and R.W. Wein. 1986. Buoyancy dynamics of floating *Typha* mats at Tintamarre Marsh, New Brunswick. Pap., 22nd An. Mtg. Canadian Botanical Assoc., Sudbury.

Holm, L., J. Pancho, J. Herberger, and D. Plunchnett. 1979. A geographical atlas of world needs. John Wiley and Sons, 391 pp.

Hotchkiss, N., and H.L. Dozier. 1949. Taxonomy and distribution of North American cattails. Am. Midl. Nat. 41:237–254.

Hutchings, M.J. 1979. Weight density relationships in ramet populations of clonal perennial herbs with special reference to the negative three-halves power low. J. Ecol. 67(1):21–34.

Keddy, P.A., and T.H. Ellis. 1985. Seedling recruitment of 11 wetland plant species along a water level gradient: shared or distinct responses? Can. J. Bot 63:1876–1879

Klots, E.B. 1966. Freshwater life. GP Putnams Sons, New York.

Krolikowska, J. 1976. Physiological effects of triazine herbicides on *Typha latifolia.* Pol. Arch. Hydrobiol. 23(2):249–259.

Laing, H.E. 1940a. Respiration of the rhizomes of *Nuphar advenum* and other water plants. Amer. J. Bot. 27:574–581.

Laing, H.E. 1940b. Respiration of the leaves of *Nuphar advenum* and *Typha latifolia.* Amer J. Bot. 27:583-586.

Laing, H.E. 1941. Effect of concentrations of oxygen and pressure upon rhizomes of some submerged plants. Bot. Gax. 102:712–724.

Leck, M.A., and K.J. Graveline. 1979. The seed bank of a fresh water tidal marsh. Am. J. Bot. 66(9):1006–1015.

Lee, D.W. 1975. Population variation and introgression in North American *Typha* spp. Taxon 24(5-6): 633–641.

Lieffers, V.J. 1983. Growth of *Typha latifolia* in boreal forest habitats, as measured by double sampling. Aquatic Bot. 15:335–348.

Linde, A.F. 1963. Results of the 1962 Horicon Marsh drawdown wetland habitat research. Ann. Prog. Rep. Job III B. Wis. Conserva. Dep. Madison, Wisconsin.

Linde, A.F., T. Janish, and D. Smith. 1976. Cattail—the significance of regrowth, phenology, and carbohydrate storage to its control and management. WI Dep. Nat. Resourc. Tech. Bul. No. 94, 27 pp.

Mallik, A.U., and R.W. Wein. 1985. Miscroscale succession of bogged paludification of a *Typha* marsh. Pap., 21st An, Mtg. Canadian Botanical Assoc., Univ. Western Ontario, Long, Ontario.

Mallik, A.U., and R.W. Wein. 1986. Response of a *Typha* marsh community to draining, flooding, and seasonal burning. Can. J. Bot. 64:2136–2143.

Marsh, L.C. 1955. The cattail story. Garden J. 114–117.

Marsh, L.C. 1962. Studies in the genus *Typha*. Ph.D. thesis, Syracuse Univ. (Libr. Congr. Card No. Mic 63-3179) Univ. Microfilm, Ann Arbor, Michigan, 126 pp.

Martin, A.C., R.C. Erickson, and J.H. Steenis. 1957. Improving duck marshes by weed control. USDA Fish and Wildlife Cir. 19.

Martin, A.C., H.S. Zim, and A.L. Belson. 1951. American wildlife and plants. McGraw-Hill Book, New York, New York, 500 pp.

McDonald, M.E. 1951. The ecology of the Pointe Mouillee Marsh, Michigan, with special reference to the biology of cattail (*Typha*). Ph.D. thesis, Univ. Michigan, Ann Arbor, Michigan (Diss, Abstr. 11:312-314).

McMillan, C. 1959. Salt tolerance within a *Typha* population. Amer. J. Bot. 46:521–529.

McNaughton, S.J. 1964. Ecotypic patterns in Typha and their significance in ecosystem integration. Univ. Microfilms, Ann Arbor, Michigan (no. 64-11 813).

McNaughton, S.J. 1966. Ecotype function in the *Typha* community type. Ecol. Monog. 36:297–324.

McNaughton, S.J. 1968. Autotoxic feedback in regulation of *Typha* population. Ecology 49:367–369.

McNaughton, S.J. 1975. R selection and K selection in *Typha*. Am. Nat. 109:251–262.

Meyer, A.M., and A. Poljakoff-Mayber. 1963. The germination of seeds. MacMillan Co., New York, New York, 236 pp.

Morinaga, T. 1926. The favorable effect of reduced oxygen supply on the germination of certain seeds. Am. J. Bot. 13:159–166.

Morton, J.F. 1975. Cattails (*Typha* spp.) weed problem or potential crop? Econ. Bot. 29:7–29.

Nelson, J.F., and R.H. Dietz. 1966. Cattail control methods in Utah. Utah Dep. Fish and Game Pub. No. 66–2, 33 pp.

Pahuja, S.S., B.S. Yadava, and S. Kumar. 1980. Chemical control of cattail *Typha augustifolia.* Indian J. Agric. Res. 14(1):13–16.

Penfound, W.T., R.F. Hall, and A.D. Hess. 1945. The spring phenology of plants in and around the reservoirs in northern Alabama with particular reference to malaria control. Ecology 26:332–352.

Pianka, E.R. 1973. Competition and niche theory. Saunders, Philadelphia, Theoretical Ecology, pp. 114–142.

Prunster, R. 1941. Germination conditions for *Typha muelleri* and its practical significance for irrigation channel maintenance. Austral. Counc. Sci. Indus. Res. J. 14:129–136.

Ristich, S.W. Fredrich, and E.H. Buckley. 1976. Transplantation of *Typha* and the distribution of vegetation and algae in a reclaimed estuarine marsh. Bul. Torrey Bot. Club. 103(4):157–164.

Roberts, E.H. 1972. Dormancy: a factor affecting seed survival in the soil. Viability of Seeds, Syracuse University Press, Syracuse, New York, pp. 321–359.

Sale, P.J.M., and R.G. Wetzel. 1983. Growth and metabolism of *Typha* species in relation to cutting treatments. Aquatic Bot. 15:321–334.

Schafer, D.E, and D.O. Chilcote. 1970. Factors influencing persistence and depletion in buried seed populations. II. The Effect of Soil Temperature and Moisture, Crop. Sci. 10:342–345.

Sculthorpe, C.D. 1967. The biology of aquatic vascular plants. St. Martin's Press, New York, New York.

Sharitz, R.R. 1974. Responses of two species of cattail to thermal effluents. Assoc. Southeast Biol. Bul. 21(2):82–83.

Shekhov, A.G. 1974. Effect of mowing times on regeneration of reed and reedmace growths. Hydrobiol. ZH. 10(3):61–65.

Sifton, H.B. 1959. The germination of light sensitive seeds of *Typha latifolia*. Can. J. Bot. 37:719–739.

Singh, S.P., and M.K. Moolani. 1973. Changes in the chemical composition of cattail induced by herbicides. Proc. All India Weed Control Semin. 3:75.

Smith, S.G. 1961. Natural hybridization and taxonomy in the genus *Typha* with particular reference to California populations. Ph.D. thesis, Univ. California, Berkeley.

Smith, S.G. 1962. Natural hybridization among five species of cattail. Am. J. Bot. 49:678.

Smith, S.G. 1967. Experimental and natural hybrids in North America *Typha* (*Typhaceae*). Am. Midl. Nat. 78:257–287.

Steenis, J.H., L.P. Smith, and H.P. Cofer. 1958. Studies on cattail management in the Northeast. Trans. 1st Wildlife Conf., Montreal, Canada, pp. 149–155.

Stodola, J. 1967. Encyclopedia of water plants. T.H.F. Pub. Jersery City, New Jersey.

Szcepanska, W. 1971. Allelopathy among the aquatic plants. Pol. Arch. Hydrobio. 18(1):17–30.

Taylor, G.J. and A.A. Crowder. 1984. Copper and nickel tolerance in *Typha latifolia* clones from contaminated and uncontaminated environments. Can J. Bot. 62:1304–1308.

Tilton, D.L., and R.H. Kadlec. 1979. The utilization of a fresh water wetland for nutrient removal from secondarily treated wastewater effluent. J. Environ. Qual. 8(3):328–334.

Timmons, F.L., et al. 1963. Control of common cattail in drainage channels and ditches. U.S. Dep. Agric. Tech. Bul. 1286, ARS, Washington, DC, 51 pp.

Uhler, F.M. 1944. Control of undesirable plants in waterfowl habitat. N. Amer. Wildl. Conf. Trans. 9:395–403.

United States Department of Agriculture. 1977. Economically important foreign weed-potential problems in the United States. Agric. Handb. No. 498, 746 pp.

United States Department of Agriculture, Agricultural Research Service. 1971. Common weeds of the United States. Dover, New York.

United States Department of the Interior. 1975. Proceedings of the National Wetland Classification and Inventory Workshop. Wildlife Management Institute, 110 pp.

United States Environmental Protection Agency. 1983a. The effects of wastewater treatment facilities on wetlands in the Midwest.

United States Environmental Protection Agency. 1983b. Environmental impact statement: Freshwater wetlands for wastewater management. 380 pp.

Van der Valk, A.G., and C.B. Davis. 1976. The seed banks of prairie glacial marshes. Can. J. Bot. 54:1832–1838.

Weller, M.W. 1975. Studies of cattail in relation to management for marsh wildlife. Iowa State J. Res. 49(4):383–412.

Wesson, G., and P.F. Waring. 1969. The role of light in germination of naturally occurring populations of buried weed seeds. J. Exp. Bot. 20:402–413.

Whigman, D.F., and R.L. Simpson. 1978. The relationship between aboveground and below ground biomass of fresh water tidal wetland macrophytes. Aquat. Bot. 5(4):355–364.

Wilcox, D.A., S.I. Apfelbaum, and R. Hiebert. 1984. Cattail invasion of sedge meadows following hydrologic disturbance in the Cowles Bog Wetland Complex, Indiana Dunes National Lakeshore. J. Soc. Wetlands Sci. 4:115–128.

Wisconsin Department of Natural Resources. 1969. Techniques of wetland management. Wisconsin Dep. Nat. Resourc. Res. Rep. 45, 156 pp.

Wisconsin Department of Natural Resources. 1971. Observations of cattails in Horicon Marsh, Wisconsin. Dep. Nat. Resourc. Res. Rep. 66, 16 pp.

Yeo, R.R. 1964. Life history of common cattail. Weed 12:284–288.

Zimmerman, J. 1987. Lecturer, University of Wisconsin -Madison. Personal communication with S. Apfelbaum, K. Montivans, June 1987.

Zohary, M. 1962. Plant life of Palestine. The Ronald Press, New York, New York.

To Contents

III.J.2.g Control and management of Sesbania (*Sesbania herbacea* (P. Mill.) McVaugh) (SEHE8)

(United States Geological Survey and Gaylord Lab, Moist Soil Advisor; edited by Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001; State distribution map by NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

This paper provides current information on the biology and control of noxious and invasive plant species that occur in wetlands.

Contents

Sesbania is an annual legume that typically grows to a height of 10 feet. It is a common nuisance species in moist soil impoundments, wetland buffers, recent restored wetlands, and agricultural fields. This plant refers wet, highly disturbed habitats and sandy sites. Optimum germination occurs later in the growing season when mudflats are exposed during periods of elevated temperatures. Although germination is late (best following late spring or summer drawdown), sesbania sometimes forms dense stands that preclude germination and growth of desirable moist-soil species. Stands of sesbania also outcompete woody plantings during the early stages of tree/shrub growth either slowing or lowering revegetation success. Seed spread to surrounding agricultural fields results in similar competition with crops and to lower yields. Longevity of seeds is great, and sporadic occurrences are common, particularly following disturbance. These sporadic occurrences increase the sesbania seed stored in the soil seedbank for future infestations.

Plant value

Sesbania produces large amounts of seeds, but value for waterfowl is poorly documented. Use of sesbania stands by green winged teals has been recorded in the Southeast, but it is undetermined whether use is related to seeds or invertebrates. Dense, robust stands tend to be avoided by waterfowl.

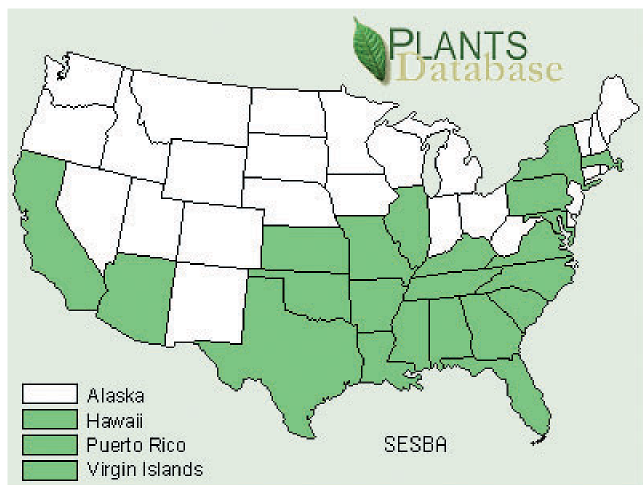
Frequency of occurrence

Problem: 5 percent cover
Severe problem: 10 percent cover

Control strategies

The control strategies for sesbania are
- agriculture
- deep disk
- deep disk then dry
- early drawdown
- late disk then flood
- mow
- plow
- herbicide
- ignore

Control of sesbania is best accomplished by creating conditions favorable for the germination of beneficial plants early in the growing season. Once established, beneficial plants can outcompete newly germinated sesbania. Therefore, control strategies should be performed early in the growing season. If early control is not possible, late disk flood often prevents reestablishment of sesbania and creates conditions favorable for fall migrating shorebirds. This can be followed by an early drawdown during the subsequent growing season.



Notes

Several scientific names have been used for *Sesbania herbacea* and can be found in older texts. They include *Sesbania exaltata* and *S. macrocarpa*. These names are considered synonyms.

III.J.2.h Control and management of cocklebur (*Xanthium strumarium* L.) (XAST)

(United States Geological Survey and Gaylord Lab, Moist Soil Advisor; edited by Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001; Seedling photograph, Bob Glennon, NRCS (currently USFWS); fruiting photograph and state distribution map by NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

This paper provides current information on the biology and control of noxious and invasive plant species that occur in wetland restoration, enhancement, and management sites.

Contents

Cocklebur is a broadleaf annual that is a common nuisance in moist soil impoundments, wetland buffers, and agricultural landscapes. Dense stands shade out more desirable vegetation or compete so effectively with moist soil plants that production of moist soil seeds is reduced. Germination requires higher soil temperatures and moist soils, thus rapid drawdowns after mid-May often result in dense stands of cocklebur. Each seedpod usually has two viable seeds that may sprout at different times in the same or different growing seasons.

Plant value

Seeds are not of value to waterfowl. The litter from cocklebur appears to provide either nutrients or substrates valuable for invertebrate production. Cocklebur is a common weed in agricultural fields and can escape to wetland restoration sites. Infestations in a wetland can become a dispersal source into an agricultural landscape where competition with crop plants can result in lower yields and increased cost in herbicide use to the landowner.

Control

The most effective control is to prevent conditions conducive to the germination of cocklebur. Thus, units with a history of cocklebur should always have slow drawdowns. Furthermore, drawdowns during periods of high ambient air temperatures should be avoided or monitored closely. Recognition of cocklebur seedlings at the 2-cotyledon stage is essential for effective control. Once cocklebur is established, the four commonly used control techniques are stress flooding, disking, mowing, and herbicides.

Stress flooding

This technique is recommended only for plants less than 6 inches in height. Cocklebur less than 3 inches in height is controlled much easier because plants must be flooded only two-thirds the plant height for 3 days or until plants begin to turn yellow. For 4- to 6-inch plants, flooding must be equal to or greater than the height of the plant and must be maintained for 57 days or until plants turn yellow. If timed correctly, stress flooding not only eliminates cocklebur, but also enhances the growth of more desirable moist soil plants.

Disking

Disking destroys the plant, but more cocklebur may germinate if soil moisture conditions become favorable following disking. If disking can be followed by irrigation, germination of cocklebur will be eliminated or at least greatly reduced. If this technique is used, care must be taken to keep the soil in a moist to saturated condition for at least 7 days to ensure germination of desirable moist soil plants. If irrigated for shorter periods, soil moisture and temperature conditions may become more favorable for germination of cocklebur.

Mowing

The timing of mowing is critical for effective control. The technique works best if performed just prior to seed set, when plants are more than 8 inches tall. Clipping the tops to within 6 inches of the ground eliminates most seed production. If performed earlier in the growing season, mowing immediately before a rain enhances the growth of seed-producing moist soil plants located below the cocklebur canopy.

Herbicide

This control method is only recommended as a last resort. Many side effects from the use of herbicides are poorly understood. Furthermore, certain herbicides have an adverse effect on desirable moist soil vegetation.

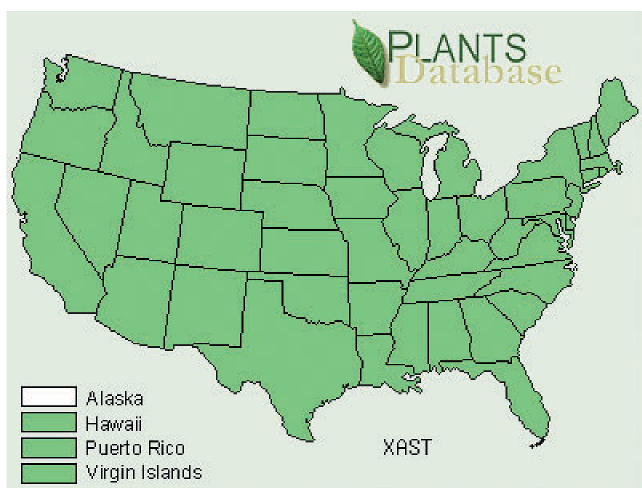
Frequency of occurrence

Problem: >10 percent as a solid block to 25 percent as scattered patches

Severe problem: >25 percent cover

Control strategies

The control strategies for cocklebur are
- agriculture
- shallow disk
- deep disk
- late disk then flood
- plow
- semi-permanent
- stress flood
- mow
- herbicide
- ignore



III.J.2.i Control and management of American lotus (*Nelumbo lutea* Willd.) (NELU)

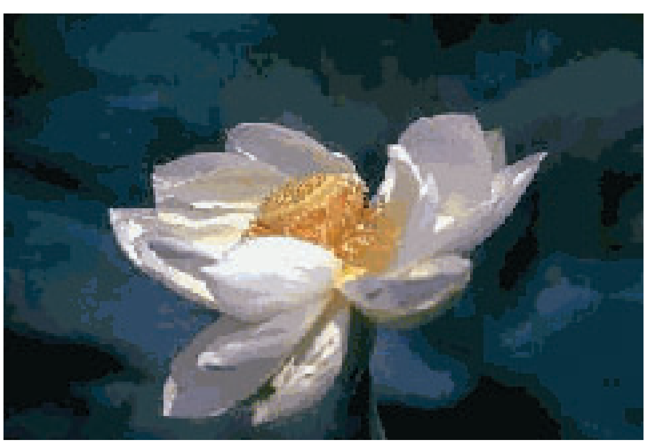
(United States Geological Survey and Gaylord Lab, Moist Soil Advisor; edited by Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001; Photographs and state distribution map by NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

This paper provides current information on the biology and control of nuisance plant species that occur in wetlands.

Contents

American lotus is a floating leaf, aquatic plant with spongy rhizomes. Adapted to sites characterized by shallow to moderate (14 feet) water depths through most of the year. Once established, this plant can withstand short periods of drought. American lotus occurs in moist soil impoundments, most commonly in ditches or low-lying areas that cannot be completely dewatered. Reproduction occurs from seeds (seeds may remain viable for more than 100 years) or vegetatively from rhizomes.

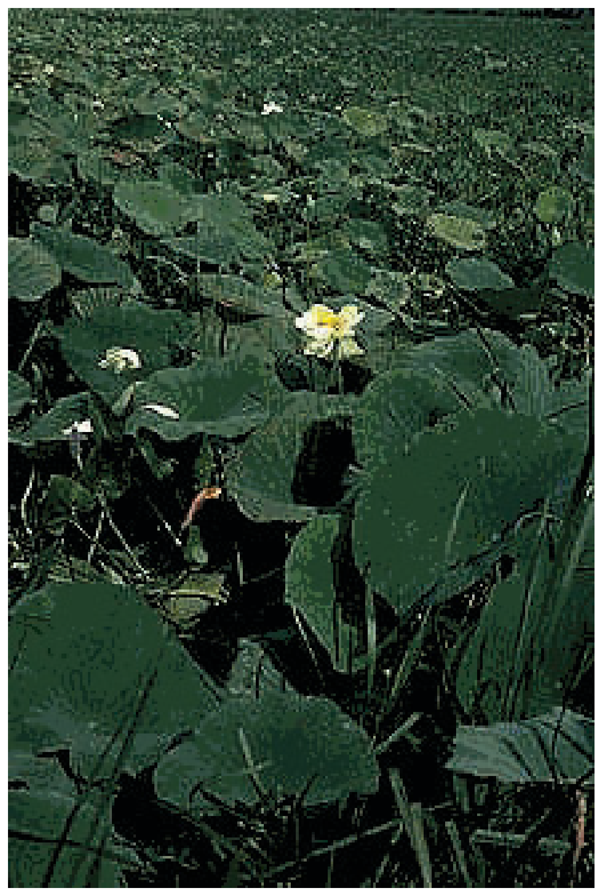


Plant value

During summer, leaves often become elevated above the water surface and serve as brood cover. Prior to plant senescence, American lotus also provides good wood duck roosting habitat. Seeds are of little value to waterfowl. Although small stands of lotus are of value in marsh systems, this plant is not considered desirable in moist soil impoundments.

Frequency of occurrence

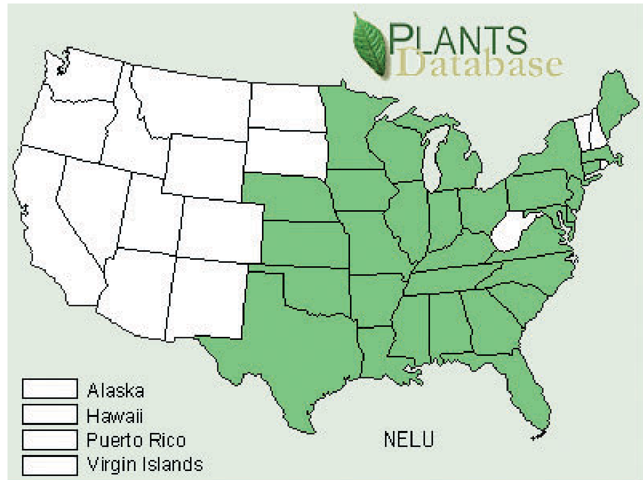
Problem: >5 percent cover
Severe problem: >15 percent cover



Control strategies

The control strategies for American lotus are
- agriculture
- deep disk
- deep disk then dry
- keep dry
- mow
- shallow disk
- herbicide
- ignore

Preventive water management schedules are the best method of controlling this species. Complete dewatering that facilitates soil drying reduces the germination potential of American lotus and promotes germination of desirable plants adapted to drier sites. If American lotus becomes well establishment, some type of mechanical treatment normally must accompany complete dewatering. In some cases combinations of treatments (i.e., deep disking and drying) may need to be performed for 2 consecutive years to accomplish adequate control.



III.J.2.j Control and management of alligatorweed (*Alternanthera philoxeroides* (Mart.) Griseb.)(ALPH)

(United States Geological Survey and Gaylord Lab, Moist Soil Advisor; edited by Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001; Photographs and state distribution map by NRCS Plant Data Center, Baton Rouge, Louisiana)

Purpose

This paper provides current information on the biology and control of nuisance plant species that occur in wetlands. Arizona, California, Florida, and South Carolina consider alligatorweed a prohibited or listed noxious weed.

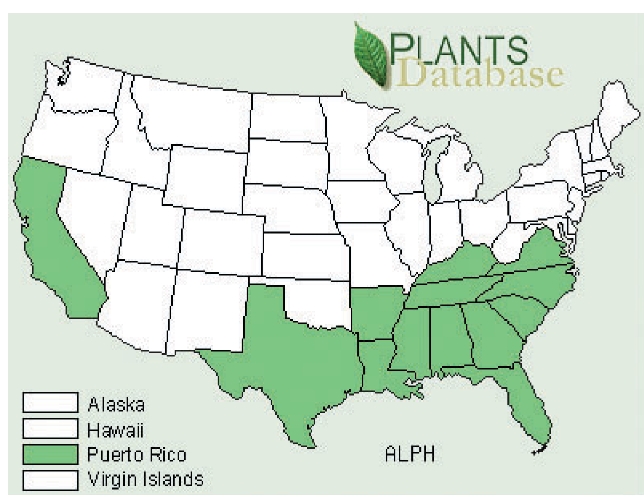
Contents

Alligatorweed is a non-native plant species with the first records of occurrence in North America from Mobile, Alabama, in 1897. This plant is assumed to have emigrated here as a stowaway in shipping ballast. It is an emersed and/or emergent, herbaceous perennial with opposite, elliptical leaves that are 0.25 to 0.75 inch long. This species tolerates a wide range of soil and water conditions: It can grow in completely terrestrial locations occurring in dry fields, or it can be free floating in ditches in fresh water (mostly) or brackish water. The plant flowers prolifically, but in the United States the populations appear to only reproduce vegetatively. In wetland situations reproduction is by fragmentation of the hollow, floating stems. They break easily at the nodes and root. It appears that land disturbance encourages the spread and establishment of alligatorweed.

The threat to wetlands that alligatorweed poses stems from its ability to form dense, interwoven, mats of vegetation over the water's surface. These mats can become several feet thick and cover an extensive surface area. The mat scrambles over native emergent aquatic vegetation and blocks sunlight penetration into the water column. This light-starves existing submerged aquatic vegetation and depresses the total dissolved oxygen. Alligatorweed grows rapidly, fragments easily, and establishes new mats downstream. The mats can block drainageways and intake pipes, alters the aquatic ecology, and may increase mosquito-breeding habitat.

Plant value

Alligatorweed has limited value for waterbirds. Although reportedly used as brood habitat, other plant species are of greater value for this purpose. Its greatest value may be for the increase of mosquito-breeding habitat.



Frequency of occurrence

Problem: If present

Severe problem: If present

Control strategies

The best control prevents the introduction, establishment, and spread of the species. It is difficult to control once it becomes established because it is adapted to a wide range of soil moisture conditions and salinities.

Mechanical methods are relatively ineffective. They are expensive, the stems fragment easily (aiding in propagation), and the site disturbance promotes spread. If mechanical control is used, the effectiveness depends on keeping the site dry for sufficient time to permit repeated mechanical disturbances (disking) to disrupt underground nutrient and energy reserves. Incomplete treatment only results in an increase of stem fragments for reestablishment.

Biological control by the importation of predators from its native range in South America has been effective in the South. These predators are not cold tolerant and loose effectiveness northward.

Herbicide applications may be necessary in dense stands and are effective in controlling alligatorweed. Herbicides should be applied with caution because these chemicals may destroy invertebrate base. Glyphosate is recommended because of its biodegradability, but it is a broad-spectrum herbicide that eliminates all vegetation.

United States
Department of
Agriculture

**Natural
Resources
Conservation
Service**

Wetland Restoration, Enhancement, and Management

Section IV Regional Issues

IV.A Restoring depressional wetland complexes for waterfowl benefits: an overview of considerations

(Michael Whited, NRCS Wetland Science Institute, Hadley, Massachusetts, December 2001)

Purpose

This paper includes the basic reasoning and strategy for restoring wetland complexes based upon research conducted in the Prairie Pothole Region (PPR) of the United States and Canada. Background information on prairie pothole classification, plant communities, and waterfowl habitat requirements is presented to facilitate an understanding of the restoration process.

Contents

Many animals require a mosaic of habitats to satisfy all their requirements for food and shelter during different life stages. Waterfowl especially are a diverse group with widely divergent requirements for survival and recruitment. Often the life habitat requirements for a species of waterfowl are best supplied by wetland clusters, or complexes, composed of a mixture of small, shallow basins and larger, deeper basins. A single wetland, even a large semipermanent wetland, cannot adequately satisfy all their requirements during different life stages (see table IV.A-1). Numerous studies have documented that the home ranges for most ducks include a large number and diversity of potholes. The term "wetland complex" refers to an assemblage of individual basins, of varying hydroperiods (i.e., wetness) in relatively close proximity to each other. No exact size specifications of a complex exist; however, the HGM Guidebook for Prairie Potholes (Lee et al. 1997) uses a 1-mile radius from the wetland being assessed to determine landscape characteristics. This is equivalent to about 2,000 acres.

Past wetland loss and degradation in North America have been severe and are implicated as a primary factor contributing to population declines of several waterfowl species. Further, most wetland complexes in the conterminous United States have been modified with respect to the spatial distribution and composition of wetland types. Disruption of wetland complexes can negatively impact many species that require several wetland types to provide the resources (i.e., nutrients, energy, nest sites) needed to successfully complete annual cycle events. For the most part, in the southern PPR (generally the part of the region where row crops dominate) wetland complexes have been eliminated by drainage and only isolated, large, semipermanent to permanent marshes remain. Because of this, the southern PPR is no longer a major breeding area for waterfowl, but rather is important primarily as a migratory stopover area.

To the waterfowl biologist in the PPR, it is the juxtaposition of different wetland types, or hydroperiod regimes, to each other and their relationship to the surrounding upland that determines waterfowl production success. Consequently, juxtaposition and interspersion of different habitats, as well as habitat quality, are important in determining use by breeding waterbirds. The decision to restore some basins, and not others, based upon the likelihood of maximizing wildlife benefits can be complicated. The following information is provided to assist in that critical, final decision.

Definitions

Because the concept of wetland complex is based on a variety of wetlands within a landscape, a thorough understanding of pothole classification is necessary. Table IV.A-1 summarizes the terminology that is commonly used when discussing types of pothole wetlands in the PPR. These terms are used throughout this document.

The Stewart and Kantrud (1971) classification system is based upon the vegetation found in the deepest zone of wetland basins. This system predates the development of a national wetland classification system (Cowardin et al. 1979). The Stewart and Kantrud (S and K) classification system classifies entire basins while the field units classified by the Cowardin et al. system are homogenous stands of vegetation. Within a landscape restoration context, the S and K system is considered more useful because it classifies entire basins, and entire basins are what are being restored (Galatowitsch and van der Valk 1994).

S and K class I, ephemeral ponds—These shallow basins are ponded temporarily and usually develop little or no wetland vegetation (Martin et al. 1953). The source of water for these areas is early season snowmelt. Undisturbed sites are a mix of tall grasses and forbs, most having a FAC or FACU wetland plant indicator status. The wetland-low prairie vegetation zone often has the highest vegetative diversity of any vegetative community on the prairies. Although ephemeral wetlands are not considered wetlands in the Cowardin et al. classification system, they provide extremely important benefits for wildlife. Because of their shallow nature, they warm early in the spring and are the first areas where wetland invertebrates provide the protein-, lipid-, and calcium-rich food required by breeding waterfowl (Batt et al. 1989). Any concept of ecological restoration for wildlife benefits should incorporate a mix of wetland water regimes, and ephemeral wetlands should be included in the mix.

S and K class II, temporary ponds—These shallow basins are ponded temporarily so that surface water is maintained for only a few weeks after the

Table IV.A–1 Comparison of systems used to classify commonly occurring depressional glaciated prairie wetlands

Stewart & Kantrud 1971 class	zones[b]	Cowardin et al. 1979 water regime modifier[c]	Martin et al. 1953 type	HGM[a]
I–Ephemeral ponds	Wetland-low prairie	—[d]	1	Precip. and snowmelt runoff Recharge
II–Temporary ponds	Wet-meadow Wetland-low prairie	Temporarily flooded —[d]	1, 2	Precip. and snowmelt runoff Recharge
III–Seasonal ponds	Shallow-marsh Wet-meadow Wetland-low prairie	Seasonally flooded Temporarily flooded —[d]	3, 4	Precip. and snowmelt runoff Recharge[e]
IV–Semi-permanent ponds	Deep-marsh Shallow-marsh Wet-meadow Wetland-low prairie	Semipermanently flooded[f] Seasonally flooded Temporarily flooded —[d]	3, 4, 5, 10, 11	Groundwater Flowthrough or discharge[g]

a HGM-All are Depressional class. Listing is dominant hydrologic source and groundwater interaction. Groundwater interaction can vary depending upon climatic conditions and season.

b Zones are listed from top to bottom, within each class, from the central zone to the outermost peripheral zone.

c Most all prairie potholes listed in this table are in the Palustrine System, Emergent class.

d The wetland-low prairie zone of Stewart and Kantrud is not considered wetland according to Cowardin et al.

e Seasonal wetlands can have significant groundwater input and can function as flowthrough wetlands, especially in the eastern part of the region (i.e., Minnesota and Iowa).

f The deep marsh zone of Stewart and Kantrud also includes wetlands with a water regime of intermittently exposed.

g Semipermanent wetlands in the eastern part of the region are dominated by Histosols, which is an indication that they are dominantly discharge wetlands.

spring snowmelt. They also can become inundated occasionally for several days after heavy rainstorms in late spring, summer, and fall. Undisturbed sites are dominated by grasses, rushes, and sedges, most having a wetland indicator status of FAC or FACW. Similar to ephemeral wetlands, they provide early season, invertebrate-rich feeding areas for breeding waterfowl. In addition, in normal to wet years, they provide habitat for breeding pairs to isolate themselves in defensible pieces of habitat.

S and K class III, seasonal ponds—These basins normally maintain surface water for an extended time in spring and early summer, but frequently are dry during late summer and fall. In the western part of the PPR, the dominant water input is snowmelt runoff and precipitation. On a gradient from west to east, shallow groundwater appears to become a more important hydrologic source for these wetlands. Undisturbed sites are dominated by emergent grasses and grass-like plants, most having a wetland indicator status of OBL and FACW. In wetter years these basins can go through an open water stage where submerged aquatic plants become common and deep marsh species (e.g., cattails) can become established. These basins can provide habitat for some species to complete a major portion of their annual lifecycle (i.e., feeding, breeding, and nesting).

S and K class IV, semipermanent ponds—These deeper and larger basins ordinarily maintain surface water throughout the spring and summer and frequently maintain surface water into the fall and winter. Groundwater is a strong component of their hydrologic source. These basins, in their normal emergent stage, are dominated by coarser and taller plants, such as bulrush and cattails. They can go through a variety of vegetative cycles, from open water to a dense cover of emergents (Weller and Spatcher 1965). Submerged or floating plants are often found throughout this zone. These wetlands provide habitat for a variety of species and are the preferred basins for diving ducks and waterfowl that nest over water (e.g., redheads). Semipermanent basins do not provide the early season, invertebrate-rich feeding areas that the shallower basins do because it takes longer for them to warm up in the spring.

Use of wetland complexes

Duck production is influenced heavily by wetland characteristics, such as quality, total area, and density of wetland basins, and size and configuration of the basins. Semipermanent and even seasonal wetlands are usually frozen when the first waterfowl arrive in the region in the spring. However, the shallow ephemeral and temporary wetlands thaw much earlier. Populations of invertebrates increase rapidly in these shallow basins, providing an early season food supply that is unavailable in other larger wetlands nearby. The abundant invertebrate populations in these shallow wetlands attract breeding pairs (Stewart and Kantrud 1973). The smaller wetlands isolate courting pairs and provide loafing sites for males near nesting hens.

During drought cycles, even the deepest parts of semipermanent wetlands are shallow enough to allow foraging by dabbling ducks, and, since seasonal wetlands are dry, semipermanent wetlands can be their principal breeding habitat during dry years. In years of normal to above normal precipitation, higher water levels in semipermanent wetlands provide overwater nesting cover of emergent vegetation required by diving ducks, but restrict feeding areas for dabbling ducks to the shallow periphery and to seasonal wetlands. Semipermanent wetlands thaw much later than shallow wetlands, but the delayed invertebrate availability coincides with the later breeding season of diving ducks (Swanson and Duebbert 1989). Hens with broods use primarily semipermanent wetlands, and to a lesser extent seasonal wetlands, until the ducklings reach flight stage. Many species of ducks move from basin to basin throughout the brood rearing season (see table IV.A-2). Later in the season, as birds go through their molt and cannot fly, they select large semipermanent and permanent wetlands with good stands of emergent vegetation that provide cover from predators. Restoration of semipermanent wetlands may have little benefit for dabbling ducks. The preferred habitat for dabbling ducks (e.g., mallards) is numerous smaller basins and perennial vegetation on surrounding uplands.

The primary reason that prairie potholes are so productive is that they do go through stages of wetting and drying, allowing nutrients to be released, thereby maintaining high levels of primary productivity. Maintenance of stable water levels has been shown to decrease primary productivity, resulting in less habitat heterogeneity and food production.

Uplands and buffers

Because many dabbling ducks, and even some diving ducks, nest in upland habitats surrounding wetlands, recruitment of waterfowl is closely tied to both terrestrial and wetland communities (table IV.A-2). Additionally, a densely vegetated buffer area provides food and shelter for brood rearing. Any pothole restoration that has waterfowl production as an objective should require at least a 50- to 75-yard-wide densely vegetated (perennials) buffer around it. Any pothole restoration that has the objective of increasing breeding success of dabbling ducks (that nest on uplands) should require restoration beyond the buffer area, well into the upland.

Another consideration is predators. Tree rows and field borders allow cover and travel lanes for mammalian predators and predatory birds (e.g., crows). Wetlands with tree rows near, or leading into, can function as waterfowl sinks by attracting waterfowl to an area where predators can actually decrease populations. Isolated depressional wetlands surrounded by narrow bands of upland vegetation may also act as waterfowl sinks because travel lanes exist in adjacent cropland and the lack of cover does not provide for hiding and escape from predators.

Table IV.A–2 Generalized habitat requirements of some common ducks in the PPR

Species	Nesting	Brood rearing	Food habits
American widgeon	Upland grassland, most within 50 yards of water.	More sedentary than most, prefer larger (> 2.5 ac) potholes.	Prefer stems and leaves of aquatic plants. Have been observed feeding on waste corn and upland grasses.
Gadwall	Upland grassland, most within 100 yards of water.	Travel up to 1 mile to preferred deep marsh and open water rearing sites.	Prefer succulent stems and leaves of aquatic plants. Will eat seeds of moist soil/mudflat plants.
Green-winged teal	Upland dense vegetation, most within 35 yards of water.	Nearby ponds, marshes, and sloughs.	Primarily vegetarians, prefer seeds on mudflats, very shallow marshes, or temporarily flooded agricultural lands.
Mallard	Upland dense cover, >24 inch high. As far as 500 yards from water. Will fly 3 to 5 miles to reach choice cover.	Will go a mile or more to preferred sites. Average only 7 days on any one pothole. Flooded whitetop, sedge, and hardstem bulrush provide preferred cover. More tolerant of open water than other dabbling ducks except pintails.	Highly adaptable, frequent varied habitats. More than any other duck, mallards use agricultural fields for feeding. Prefer small grains and corn. Rarely dine on soybeans.
Pintail	Upland sparse cover, even stubble fields. Most within 100 yards of water, some as far as a mile or more.	The greatest transients of all ducks. Often do not occupy a single pothole for > 14 days.	Make great use of cereal grains; can damage crops by feeding and trampling, especially swathed grain. Make little use of waste corn. Feed extensively on seeds of moist soil plants (smartweed, millets).

Table IV.A–2 Generalized habitat requirements of some common ducks in the PPR—Continued

Species	Nesting	Brood rearing	Food habits
Bluewinged teal	Upland grass, hayland, buckbrush, sedge meadows. Most within 50 yards of water.	Broods travel 100 to 1,600 yards to rearing areas.	Vegetative parts of aquatic plants, seeds of mudfalt plants (smartweed, millets), and fourth of diet is invertebrates.
Northern shoveler	Upland grass, hayfields. Most 25 to 75 yards from water.	Broods seldom remain on any pothole more than 7 to 10 days.	Feeds actively in deep and shallow water. Feeds on surface plankton, invertebrates, and seeds of aquatic and emergent vegetation.
Canvasback	Over open water 6 to 24 inches deep in dense, coarse, emergent vegetation (cattails).	Prefer the most open, largest, and deepest potholes.	Feed on plants (80%) and invertebrates (20%). Sago pondweed an important dietary component.
Redhead	Most over open water, some on uplands. Over 85% within 50 yards of water. Prefer emergent vegetation of large marshes and semi-permanent potholes.	Open water and deep marsh.	Feed more extensively on aquatic plants and less on animal life than other diving ducks. Pondweeds single most important dietary component. Juveniles prefer whitetop seeds.
Lesser scaup	More prone than other diving ducks to nest on upland areas adjacent to lakes and deeper potholes. Nest on sedge, juncus beds, and mixed prairie. Most within 50 yards of water.	Similar to other diving ducks; open water and deep marsh. Large, but poorly defined home range.	Feed in deeper water than do other diving ducks. Animal life (snails and bottom dwellers) makes up 90% of summer foods on breeding grounds.
Ruddy duck	Large and small marshes, emergent vegetation, near shore.	More sedentary than any other prairie duck species.	Primarily vegetarians, secondarily consumers of animal life. More prone to feed in small waterbodies than scaups and canvasbacks.

Source: Bellrose, F.C. 1980. Ducks, geese and swans of North America, 3rd ed. Wildlife Management Institute, Stackpole Books, Harrisburg, Pennsylvania.

Wetland densities

Tables IV.A–3 and IV.A–4 summarize most of the existing data on densities and classes that exist on the landscape in the PPR. It is apparent that the smaller, shallower wetlands greatly outnumber semipermanent wetlands on the landscape. Ecological restoration should attempt to mimic these densities and classes.

Table IV.A–4 Wetland classes in the Prairie Pothole Region

Wetland size	Location	Source
50% < 0.5 acre	Saskatchewan	Adams 1988
60% < 1 acre	Alberta	Merriam 1978
66% < 0.5 acre	U.S. - PPR	NRI, NRCS 1995
95% temporary & seasonal	Glac. Plain (ND and SD)	Reynolds et al. 1997
85% temporary & seasonal	Prairie Coteau (SD)	Reynolds et al. 1997
92% temporary & seasonal	Missouri Coteau (ND and SD)	Reynolds et al. 1997

Notes:
- There are more semipermanent wetlands in the Coteau areas.
- There are many more seasonal and temporary (than semipermanent) wetlands throughout the PPR.
- Therefore, ecological restoration should focus on the smaller, "drier" wetlands.

Table IV.A–3 Wetland basins on a landscape basis in the Prairie Pothole Region

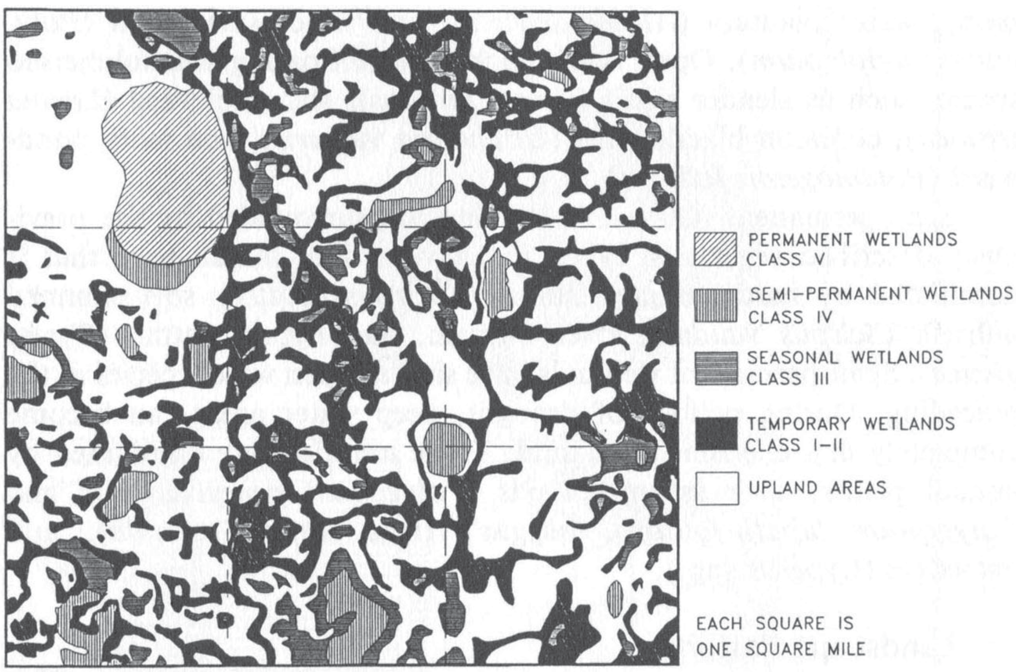
Density (/mi^2)*	Density (/km^2)*	Landscape	Location	Source
23–34	9–13		North Dakota	Cowardin et al. 1981
155	60	Moraine	Canada	Harmon 1970
233	90	Moraine	Canada	Munro 1963
52–75	20–28	Moraine	SW Manitoba	Adams 1988
52	20	Moraine	Central Saskatchewan	Adams 1988
41	16	Moraine	Missouri Coteau (ND&SD)	Gilbert 2000
33–41	24-33	Moraine	Missouri Coteau (ND&SD)	Reynolds et al. 1997
21–27	8-10	Moraine	Prairie Coteau (SD)	Reynolds et al. 1997
36	14	Moraine	Prairie Coteau (SD)	Gilbert 2000
36	14	Glaciated plains	ND and SD	Gilbert 2000
13	5	Glaciated plains	IA and MN	Gilbert 2000
36–50	14-19	Glaciated plains	ND and SD	Reynolds et al. 1997

* Density = Number of basins per area.

Notes:
- The number of basins (density) is similar in Missouri Coteau and Glaciated Plains.
- The classes of basins are different by landscape; i.e., more semipermanent basins in Coteau areas.
- The density of basins in the Prairie Coteau is lower than in the Missouri Coteau.
- The density of basins is significantly lower in the Glaciated Plains of Iowa and Minnesota. This is probably explained by two factors: historic loss in the more intensively farmed area and differences in NWI methodology and mapping between the two FWS/NWI regions.

Use of soils to determine historic complexes The use of soil types (series) is an excellent way to determine what types of wetlands existed on the landscape before conversion (see table IV.A–5). Research conducted by North Dakota State University (Arndt and Richardson 1988; Richardson et al. 1994), USGS (Winter and Rosenberry 1995), and others have correlated wetland classes with soil series. By determining the conditions present prior to agricultural development on the landscape, ecological restoration can be geared to reproducing historic wetland complexes. Figure IV.A-1 is an example of using soils information to gain an understanding of what the wetland complex was before agricultural conversion.

Figure IV.A–1 The predrainage extent of wetlands in a 9-square-mile area of Wright County, Iowa, as estimated from county soil maps*



* Copied from Galatowitsch and van der Valk (1994), figure 2.2, page 16.

Wetland classes correspond to soil map units as follows:

Wetland class	Soil map units
Class V	Open water
Class IV	Palms (211), Okoboji muck (90)
Class III	Okoboji silty clay loam (6)
Class I-II	Webster (107), Canisteo (507), Harps (95), Kossuth (388)

Table IV.A–5 Hydric soil series of the eastern PPR associated with Prairie Pothole Wetlands in loamy and clayey textured materials (modified from Galatowitsch and van der Valk. 1996)

S & K plant community	Soil taxonomy	Drainage class	Soil series	Local landscape	Ponding depth (as per soils db)
Low prairie	Endoaquolls	P	Delft, Flom, Kossuth, Letri (epi), Madelia, Perella (epi), Romnell, Rushmore, Webster	Swales (open depressions) & rims of depressions	None
	Vertic Epiaquolls & Epiaquerts	P	Beauford, Brownton, Fulda, Marna, Waldorf		None
Low prairie to wet meadow	Calciaquolls	P	Badus, Colvin, Harps, Vallers	Rims of depressions & subtle rises on flats	None
Low prairie to wet meadow	Endoaquolls with shallow calcar-eous horizons	P & VP	Canisteo, Jeffers, Spicer	Rims of depressions & subtle rises on flats	+1
Wet meadow	Argialbolls	P	Tetonka, Tonka	Shallow depressions	+1
Wet meadow to shallow marsh	Vertic Epiaquolls	P & VP	Dovray, Lura	Shallow depressions	+1
Wet meadow to shallow marsh	Argialbolls	P & VP	Barbert, Rolfe	Shallow depressions	+1
Wet meadow to shallow marsh	Endoaquolls	P & VP	Calcousta, Lanyon, Quam, Wacousta	Depressions	+1
Shallow marsh to deep marsh		P & VP	Afton, Baltic, Glencoe, Knoke, Okoboji, Oldham, Parnell, Worthing	Depressions	+3

Considerations

If the wetland(s) to be restored is (are) ephemeral or temporary:

- Is there a permanent source of water within 2 miles?
  If not, waterfowl benefits may be negligible.
- Is there a permanent source of water within 1 mile?
  If not, waterfowl benefits may be low.
- If there is no permanent source of water within 1 to 2 miles, are there any semipermanent wetlands within 1 mile?
  If not, waterfowl benefits may be negligible.
- Is the wetland to be restored in an area of medium to high density of wetlands (30+ wetlands per mile^2)?
  If not, waterfowl benefits may be low.
- Is the wetland to be restored part of a complex of other wetlands and restored uplands?
  If not, waterfowl nesting success may be minimal.

If the wetland(s) to be restored is (are) semipermanent:

- Does the area (1 mile radius from proposed restoration) have more than 5 to 10 percent of existing wetland basins semipermanent or wetter?
  If the area does, than additional large wetlands are not necessary for waterfowl.
- Will the proposed restoration have adjacent upland restored?
  If not, the basin may only provide migratory habitat.
- Is the proposed restoration part of a landscape-scale restoration involving additional smaller wetlands and upland habitats?
  If not, waterfowl benefits may be negligible.
- Is the wetland to be restored part of a complex of other wetlands and restored uplands?
  If not, waterfowl nesting success may be minimal.

Recommendations

- Restoration of small basins should be within 1 mile of semipermanent or permanent wetlands.
- No more than 5 to 10 percent of the basins in an area should be semipermanent or wetter.
- Stable water levels negatively impact the maintenance of primary production, which is the key to lush vegetation and invertebrate populations.
- Water levels need to be drawn down and flooded to mimic natural drought and wet cycles to keep productivity high.
- Wetlands should be in an area of moderate to high density of wetlands, at least 30 per square mile.
- Soil types (soil series) can be used as predictors of historic conditions.
- Restoration of semipermanent basins should have upland buffers at least 50 yards wide to provide habitat for diving ducks that use uplands.
- Restored wetlands should not have tree rows leading into or along the edge of them because the trees provide habitat and travel routes for predatory mammals and birds.
- Landscape restoration should consider restoration of a variety of wetland types, including ephemeral wetlands, along with uplands. Rather than focusing on wetland restoration, the focus should be on restoring an ecologically functioning block of land.

References

Adams, G.D. 1988. Wetlands of the prairies in Canada. *In* Wetlands of Canada, National Wetlands Working Group, Canadian Comm. Ecol. Land Class., Canadian Wildl. Serv., Environ. Canada, Ecol. Land Class. Series, No. 24, pp. 157–198.

Adams, G.D., and R.C. Hutchinson. 1976. Land capability for wildlife-waterfowl. Canada Land Inventory Map No. 62K-Riding Mountain, 1:250,000 map, Canada Dep. Reg. Econ. Expansion, Ottawa, Ontario.

Arndt, J.L., and J.L. Richardson. 1988. Hydrology, salinity and hydric soil development in a North Dakota prairie-pothole wetland system. Wetlands 8:93-108.

Batt, B.D.J., M.G. Anderson, C.D. Anderson, and F.D. Caswell. 1989. The use of prairie potholes by North American Ducks. In A.G. van der Valk (ed.), Northern Prairie Wetlands, Iowa State Univ. Press, Ames, Iowa.

Bellrose, F.C. 1980. Ducks, geese and swans of North America, 3rd ed. Wildlife Mgt. Inst., Stackpole Books, Harrisburg, Pennsylvania.

Cowardin, L.M., V. Carter, F.C. Golet, and E.T. LaRoe. 1979. Classification of wetlands and deepwater habitats of the United States. FWS/OBS-79/31.

Cowardin, L.M., D.S. Gillmer, and L.W. Mechlin. 1981. Characteristics of central North Dakota wetlands determined from sample aerial photographs and ground study. Wildlife Soc. Bul. 9(4):280–288.

Galatowitsch, S.M., and A.G. van der Valk. 1994. Restoring prairie wetlands: an ecological approach. Iowa State Univ. Press, Ames, Iowa.

Galatowitsch, S.M., and A.G. van der Valk. 1996. Characteristics of recently restored wetlands in the prairie pothole region. Wetlands 16(1):75–83.

Gilbert, M.C. 2000. Unpublished data. U.S.A.C.E., Omaha, Nebraska.

Harmon, K.W. 1970. Prairie potholes. National Parks and Conservation Mag. (Canada) 45(3):25–28.

Lee, L.C., M.M. Brinson, W.J. Kleindl, P.M. Whited, M. Gilbert, W.L. Nutter, M.C. Rains, D.F. Whigham, and D. DeWald. 1997. Operational draft guidebook for the hydrogeomorphic assessment of temporary and seasonal prairie pothole wetlands.

Martin, M.C., N. Hotchkiss, F.M. Uhler, and W.S. Bourn. 1953. Classification of wetlands of the United States. U.S. Fish and Wildl. Serv., Spec. Sci. Rep. Wildl. No. 20.

Merriam, G. 1978. Changes in aspen parkland habitats bordering Alberta sloughs. Canadian Field Naturalist 92(2):109–122.

Munro, D.A. 1963. Ducks and the Great Plains wetlands. Canadian Audubon Magazine, September-October 1963, 8 pp.

Reynolds, R.E., D.R. Cohan, and C.R. Loesch. 1997. Wetlands of North and South Dakota. Jamestown, ND: Northern Prairie Wildl. Res. Ctr. homepage, http://www.npwrc.usgs.gov/resource/distr/others/wetstats/wetstats.htm (Ver. 01OCT97).

Richardson, J.L., J.L. Arndt, and J. Freeland. 1994. Wetland soils of the prairie potholes. Advances in Agronomy 52:121–71.

Shaw, S.P., and C.G. Fredine. 1956. Wetlands of the United States. U.S. Fish Wildl. Serv. Circ. 39, 67 pp.

Stewart, R.E., and H.A. Kantrud. 1971. Classification of natural ponds and lakes in the glaciated prairie region. U.S. Dep. Int., Fish and Wildlife Serv., Bur. Sport Fish. and Wildl., Resource Pub. 92.

Stewart, R.E., and H.A. Kantrud. 1973. Ecological distribution of breeding waterfowl populations in North Dakota. J. Wildl. Mgt. 54:433–437.

Swanson, G.A., and H.F. Duebbert. 1989. Wetland habitat of waterfowl in the prairie pothole region. In. A.G. van der Valk (ed.), Northern Prairie Wetlands, Iowa State Univ. Press, Ames, Iowa.

United States Department of Agriculture, Natural Resources Conservation Service. 1995. National Resources Inventory.

Weller, M.W., and C.E. Spatcher. 1965. The role of habitat in the distribution and abundance of marsh birds. Iowa State Univ. Agric. and Home Econ. Exp. Sta. Spec. Rep. No. 43.

Winter, T.C., and D.O. Rosenberry. 1995. The interaction of ground water with prairie pothole wetlands in the Cottonwood Lake Area, East-Central North Dakota, 1979–1990. Wetlands 15(3):193–211.

IV.B Sedimentation of depressional wetlands in agricultural settings

(P. Michael Whited, NRCS Wetland Science Institute, Hadley, Massachusetts, December 2001)

Introduction

Wetland loss has been estimated at more than 50 percent in the conterminous United States (Dahl 2000) and as high as 90 percent in many agricultural areas (e.g., Iowa). Factors affecting the water volume in the remaining wetlands, however, are less obvious and not often considered. Currently, accumulation of sediment may be having the most insidious impact on depressional wetlands.

Sediment is the most important pollutant of surface water in the United States (Ellis 1936), and the greatest source of sediment is erosion of agricultural lands (Long 1991, Wayland 1993). Sediment deposition is a natural geologic process that is maintained over thousands of years. Sediment retention by wetlands is often touted as a water quality benefit (Boto and Patrick 1978). However, accelerated sedimentation may have the most negative impact on depressional wetlands because the shallower basins that result store less water. Lower potential water storage in wetlands has negative impacts on biodiversity, water supply, and can result in increased runoff that exacerbates flooding.

Many depressional wetlands are embedded within an agricultural landscape where cultivation of wetland catchment areas (i.e., the area that contributes surface runoff to the wetland basin) has greatly altered surface runoff dynamics and hydrologic inputs to ground water. Grasslands and woodlands that once protected soils from erosion and moderated surface runoff have been converted to cropland. Consequently, wetlands in agricultural fields receive significantly more surface runoff containing sediment than occurred before agricultural conversion (Grue et al. 1986, Neely and Baker 1989, Euliss and Mushet 1996, Gleason 1996, Luo et al. 1997). Spatial position and morphology of depressional wetlands in agricultural fields make them highly vulnerable to sedimentation (Luo et al. 1997, Martin and Hartman 1987, Kantrud et al. 1989, Gleason 2001).

The impact of sediment on depressional wetlands has been shown to decrease water storage volume, alter hydroperiods, lower soil organic matter, reduce plant richness and primary productivity, and negatively impact wildlife (Freeland and Richardson 1996, Gleason 2001, Jurik et al. 1994, Luo et al. 1997, van der Valk and Pederson 1989).

Restoration projects involving depressional wetlands require consideration of sedimentation and its impacts on wetland functions. It may be necessary to remove anthropogenic sediment to restore appropriate hydrology, native plant communities, and biogeochemical functions. The key to maximizing life spans of restored depressional wetlands is reducing sediment inputs.

Processes

Particulates are transported into depressional wetlands from several sources. Sources include dry deposition and precipitation from the atmosphere, overland flow from adjacent uplands, and occasional overflows connecting wetlands during wet periods of high storage (Adomaitis et al. 1967, Grue et al. 1989, Luo et al. 1997, Gleason 2001). Atmospheric sources are assumed to account for a relatively small amount of the total quantity of particulates that typically impact depressional wetlands. However, in areas of intense agriculture, atmospheric deposits of particulates in wetlands may be significant (Adomaitis et al. 1967, Frankforter and Frenzel 1995).

Most depressional wetlands are surficially closed basins that lack integrated drainage networks (Richardson et al. 1994). Thus, wetland sediment inputs are derived primarily from wind and water erosion of upland soils in adjacent areas. Tillage has greatly altered the surface hydrologic dynamics of wetland catchments; conventional tillage increases erosion rates and surface runoff relative to grassland landscapes (Gleason 1996; Euliss and Mushet 1996; Luo et al. 1997). Adomaitis et al. (1967) demonstrated that the aeolian mixture of snow and soil (snirt) in wetlands surrounded by fields without vegetation accumulated at twice the rate as in wetlands surrounded by fields with vegetation. Similarly, Martin and Hartman (1987) found that the flux of inorganic sediment into wetlands with cultivated catchments occurred at nearly twice the rate of wetlands with native grassland catchments. Dieter (1991) demonstrated that turbidity was highest in tilled (i.e., wetland and catchment areas tilled) than in untilled and partly tilled (i.e., portions of the basin tilled with a buffer strip of vegetation separating the basin and catchment area) wetlands. In the playa wetlands of Texas, Luo et al. (1997) found that wetlands in cultivated watersheds had lost nearly all of their original volume due to filling by sediment, whereas comparable sites in rangeland watersheds lost only about a third of their original volume. Both Dryer et al. (1996) and Gleason (2001) found that sediment accretion rates were up to three times higher in cropped watersheds than in grassland watersheds in the prairie pothole region (PPR) of the United States. A conclusion common to all these studies is that wetlands in agricultural landscapes have shorter topographical lives than wetlands in grassland landscapes.

Impacts

Excessive sediment input potentially alters aquatic food webs as well as basic wetland functions related to water quality improvement, nutrient cycling, and other biogeochemical processes that transform and sequester pollutants. Moreover, erosional sediment can fill wetlands either as a single catastrophic event or gradually; basins totally filled with sediment provide no natural wetland functions of benefit to society.

The extent of sediment delivery to the wetland from culturally accelerated sources decreases, or even eliminates, water storage volume, alters hydroperiods; reduces plant richness, emergence, and germination; and encourages monodominant plant communities (e.g., cattails and reed canarygrass) (Luo et al. 1997, Gleason and Euliss 1998, Jurik et al. 1994, Wang et al. 1994).

Imported elements and compounds that are attached to the sediment particles, such as phosphorus, are also deposited in the wetland. This in turn, affects the capacity of the basin to sustain biogeochemical processes over the long-term.

Effects on hydrologic functions

The effect of wetland sediment on groundwater hydrology is unknown, but the alteration of the ratio of surface water to groundwater hydrology in wetlands is obvious. As the native landscape was converted to cropland, the runoff dynamics of the entire landscape were changed. Surface runoff from snowmelt and storms during presettlement times was moderated by native vegetation, dampening the effect of runoff and increasing the time available for infiltration. Conversion of native grassland to cropland has increased the intensity of runoff events and decreased the time available for infiltration. The unusually high variance in water level fluctuations in PPR wetlands in agricultural landscapes found by Euliss and Mushet (1996) was attributed to higher runoff potential of cropland versus grassland. Further, modifications to presettlement surface runoff dynamics result from an extensive road system in many agricultural regions, with roads often occurring in both north-south and east-west orientation at roughly 1.61 km (1 mile) intervals. Most of these roads are elevated, and many lack adequate culvert systems to pass water through traditional paths of conveyance. As a result, the sheetflow dynamics have been severely altered (Luo et al. 1997) and the importance of surface flow has greatly increased in recent times. Increased surface flow can exacerbate flooding as was noted by Miller and Nudds (1996) who related intensity of floods in the Mississippi River Valley to landscape change involving conversion of grassland to cropland in the prairies.

Aside from altering the natural ratio of ground to surface water input into wetlands, wetland sedimentation may have altered local groundwater flow patterns. Precipitation that was once lost through evapotranspiration or infiltration to ground water before entering wetlands in grassland catchments, may now enter wetlands via spates of surface runoff from tilled catchments. These surface runoff spates may transport sediment, nutrients, and other pollutants into wetlands (Goldsborough and Crumpton 1998). In addition to the alteration of hydrologic inputs, the loss of basin volume from siltation reduces the water storage capacity and flood attenuation benefits of wetlands (Ludden et al. 1983, Luo et al. 1997, Gleason 2001).

Effects on primary production

Anthropogenic sedimentation can suppress primary production, reduce dissolved oxygen levels, and alter natural food chain interactions. Increased sediment in the water column generally reduces the depth of the photosynthetic zone and hence reduces the light available for primary production by aquatic macrophytes and algae (Ellis 1936, Dieter 1991). As sediment falls out of suspension, deposition may be adequate to bury algae, submergent plants, and seedbanks (Rybicki and Carter 1986, Hartleb et al. 1993, Jurik et al. 1994, Wang et al. 1994). Jurik et al., Wang et al., and Gleason (2001) demonstrated that sediment depths of as little as 0.25 to 0.5 centimeter could significantly reduce species richness, emergence, and germination of wetland macrophytes.

The magnitude and timing of anthropogenically accelerated sedimentation influence structure and recolonization of plant communities in wetlands. Under natural conditions, plant communities in wetlands are dynamic and undergo cyclic changes in response to short- and long-term water-level fluctuations and salinity. Four prairie pothole wetland cyclic conditions were identified by van der Valk and Davis (1976): dry marsh, regenerating marsh, degenerating marsh, and lake. During the dry marsh or drawdown phase, sediment and seedbanks are exposed and mudflat annuals and emergent plant species germinate and recolonize the wetland. Since recolonization is dependent on viable seedbanks, the covering of seedbanks with sediment has can potentially impede the recolonization process (Jurik et al. 1994). Additionally, loss of wetland volume from accelerated sedimentation makes wetlands shallower. This allows monodominant stands of cattails, normally restricted to water depths of less than 60 centimeters (Bellrose and Brown 1941), to expand. Such stands contribute little to biological richness and exacerbate problems with agricultural interests. For example, they provide roost sites for blackbirds that depredate cereal crops (Linz et al. 1996).

Effects on aquatic invertebrates

Any suppression of primary production from sedimentation would be expected to negatively impact wetland invertebrates. The loss of standing vegetative structure generally makes wetlands less productive of invertebrates (Euliss and Grodhaus 1987). Recent studies stressing the nutritional value of algae to invertebrates (Neill and Cornwell 1992) suggest that loss of algal biomass also would make wetlands less productive of invertebrates. Sedimentation may cause a shift from herbiverous and filter feeding species (e.g., midges, zooplankters, mayflies) to sediment burrowing species (e.g., aquatic worms) (Adamus 1996). Thin layers of sediment covering the substrate also affect the success of many benthic species.

Effects on wildlife

An important function of wetlands is to provide wildlife habitat. Alteration of vegetative cover and aquatic invertebrate communities directly impact these organisms and other wetland wildlife that use them for food or cover. Aquatic invertebrates are important dietary items of waterfowl (Krapu 1974, Swanson and Duebbert 1989, Euliss and Harris 1987) and other wetland-dependent birds (Reeder 1951). The impact of sedimentation on wetland wildlife is most likely indirect, involving habitat changes in response to siltation events. An impact on plant communities is the reduction of runoff associated with restoring the surrounding upland to perennial plant cover. This change in runoff dynamics in conjunction with a lessened pool depth due to sedimentation can result in restored wetlands not supporting wetland plant communities (van der Kamp et al. 1999). Cumulatively, changes in water regimes on a landscape scale may dramatically impact wetland wildlife.

Effects on water quality functions

The water quality functions wetlands provide are dependent upon interactions between vegetation, substrates, and microbial populations (Hammer 1992). Wetland soils are the primary media wherein microbial mediated transformation of nutrients and storage of pollutants occur. The most active sites of chemical transformations are the thin aerated zones at the soil-water interface and the thin aerobic zone surrounding the roots of vascular plants (Hammer 1992, Mitsch and Gosselink 1993). For agrichemicals that require biological or chemical transformation for solubilization and subsequent removal, there is potential for sediment burial. The chemicals can then accumulate in the system (Neely and Baker 1989).

Although the impact of sedimentation on specific processes involved with improving water quality in wetlands is poorly understand (Adamus and Brandt 1990), the indirect impact sediment exerts on water quality through its influence on hydrophytes, organic exchange substrates, and microbial populations is better understood. Reduction of light available for photosynthesis due to turbidity and the burial of macrophyte seedbanks are obviously negative impacts of excessive sediment entering wetlands from adjacent fields. Aquatic macrophytes and algae are important in the uptake, short-term storage, and cycling of nutrients in wetlands. Negative impacts on plants from sediment may alter water quality functions. Increased input of allochthonous inorganic matter to wetlands (Martin and Hartman 1987, Gleason 1996) reduces the availability of organic exchange surfaces important for sorption of contaminants, especially on the thin aerobic zone at the soil-water interface. While the impact of sedimentation on microbes has not been studied (Adamus and Brandt 1990), sediment fallout may cover microbes or organic matter needed for microbial processes, or alter redox profiles important in the performance of water quality processes.

Finally, the ability of wetlands to remove and retain sediment is a basic concept of improved water quality, but many depressional wetlands are closed systems that can totally fill with sediment and hence lose their capacity to function properly. The tradeoff between the importance of sediment removal as a water quality benefit and maintaining the topographic life of wetland basins clearly needs to be integrated into management strategies of wetlands.

Assessment of sedimentation

Direct evidence of retained sediment is the best qualitative indicator. Occasionally, layers of leaves or vegetation are buried under sediment layers, but such rates of deposition are infrequent in most small watersheds. Another indicator of sediment deposition is the presence of a buried A horizon in the soil profile or lighter colored depositional material on the soil surface. Luo et al (1997) used the depth to the soil Bt (i.e., claypan) layer in playa wetlands as an indication of accelerated sedimentation. The thickness of the A horizon in prairie pothole wetland soils has been used by others as an indicator of sediment (Lee et al. 1997).

Sediment may also be "modeled" using an erosion prediction model, such as RUSLE (Renard et al. 1997). Although RUSLE does not model sediment delivery, it is a relatively safe assumption to expect catchment erosion to be directly related to sedimentation. Caution needs to be taken when using any predictive approach because sedimentation rates are dependent upon land use over time. For example, a restored wetland in a reseeded prairie may not appear to be receiving much sediment today, but historically it may have received large quantities of sediment (Gleason 2001).

Recommendations

Assess sediment inputs and impacts

An assessment of past and present sedimentation and its impacts on wetland functions is necessary for proposed restorations. Minimally, the assessment should include estimates of the depth of sediment and the resultant loss of water storage volume in depressional wetlands.

- Will it be necessary to remove sediment or increase wetland pool depths with structures to restore appropriate hydrology and hydrophytic plant communities?
- Has sediment, in conjunction with drainage and cultivation (Weinhold and van der Valk 1988), effectively depleted the seedbank so that active revegetation is necessary to restore desired native plant communities?
- Will any resultant loss of water storage and increase in nutrients result in undesirable monodominant stands of vegetation (e.g., cattail, reed canarygrass)?

Methods:

- Evaluate historic erosion/sedimentation from water using such tools as RUSLE.
- Evaluate historic erosion/sedimentation from wind (where appropriate) using tools, such as WEQ or WEPS.
- Investigate soil profiles for evidence of sedimentation (e.g., buried A horizon, thickness of A horizons/depth to Bt horizon, buried plant materials, lighter colored or different textured overburden, calcareous overwash, and elevated phosphorus levels).
- Identify current sediment inputs using accepted soil erosion prediction technology.

- Determine historic water regime of wetland (using soils as indicators of water regime, water budgets, catchment/basin ratios).
- Determine if sedimentation has changed the historic water storage (survey cross sections of present surface versus presediment surface).

Reduce sediment inputs

Evaluate the effectiveness of alternative agricultural practices to reduce erosion from cropland and sedimentation of wetlands. Depressional wetlands in agricultural settings should not be restored without protective, perennially vegetated buffers around them and best management practices on surrounding cropland to minimize ongoing sedimentation. Vegetative buffer strips are frequently used and have been shown effective at reducing nonpoint source pollutants, including sediment, from adjacent habitats (Castelle et al. 1992). The semiarid Great Plains undergoes long periods of drought followed by long periods of abundant rainfall. These wet/dry cycles can persist for 10 to 20 years (Duvick and Blasing 1981, Karl and Riebsame 1984). During periods of severe drought, most wetlands go dry during summer and many remain dry throughout the drought years. Buffer strips established to protect wetlands during a dry cycle may become submerged and ineffective in reducing sediment input in wetlands during the wet cycle (Gleason 1996). Establishing permanent vegetative cover in a depressional wetland catchment basin and its associated upland is the best practice for reducing sedimentation rates.

When constructing a road or any drainage ditch in areas of depressional wetlands, a barrier should be constructed to prevent sediment from being deposited into the basin. Vegetative barriers (NRCS Practice Standard 601) can be effective, especially in concentrated flow areas. Fields should not be listed toward a wetland. In some areas of the United States, wetlands collect irrigation water and its accompanying sediment (e.g., playas, rainwater basins). When fields are irrigated, care should be taken to prevent sediment runoff into a wetland. The use of sprinkler irrigation greatly increases the effectiveness of irrigation and reduces runoff.

Methods:

- Do not restore depressional wetlands without perennially vegetated buffer zones of a minimum 100-foot width, the wider the better.
- Consider broad, climatic shifts and maximum/minimum pool depths when establishing buffer areas in semiarid climates
- Establish perennial cover on adjacent uplands, or implement soil conservation practices on adjacent agricultural lands
- Convert adjacent irrigated fields to sprinkler type irrigation. (At a minimum do not run irrigation rows up and down hill into basins without some sort of sediment trapping mechanism.)
- Use silt trapping fences on contours of slopes, and check dams in concentrated flow areas when construction activities are occurring.

Restore pool depths

Methods to restore drained or nondrained wetlands that are silted in and have lost their original wetland volume need to be evaluated within the context of economics and their post restoration potential to provide targeted functions. Excavation of sediment and/or increasing the water depth with water control structures to restore historic hydrology may have the same effect of restoring water depth, but unfortunately the economic cost versus gain in wetland functions are not known.

Once it has been determined that pool depths do not reflect historic volumes and the decision has been made to take action to rectify the situation, one of two approaches can be used. The pool depth can be increased artificially using structural measures (e.g., dikes), or the anthropogenic sediment can be excavated.

Excavate sediment

Advantages:

- Minimizes flooding of adjacent land
- May expose historic seedbank and reduce need for active revegetation
- Removes nutrient/contaminant laden topsoil
- Removes some weedy plant species
- Most likely to restore entire suite of functions by mimicking natural condition

Disadvantages:

- More expensive than installing structural measures
- Overexcavation can have negative consequences for wildlife and biogeochemical functions by creating steep sided, open water wetlands

Install a dike

Advantages:
- Less expensive than excavating sediment

Disadvantages:
- May flood adjacent land
- Dikes may leak; higher water levels may result in seepage loss through more permeable adjacent upland soils
- Natural vegetation less successful
- Elevated nutrient level in sediment results in monodominant or weedy plant stands (e.g., cattails, reed canarygrass)
- Restores water storage function, but may negatively impact other functions

Summary

Many depressional wetlands are embedded in agricultural landscapes where tillage of their catchment areas facilitates increased surface runoff and sediment inputs relative to a grassland condition. Erosional sediment from anthropogenic sources can greatly shorten the topographic life of depressional wetlands. Luo et al. (1997) in their study of playa wetlands in Texas states that "if sedimentation continues as it has for the past 60 years, crop playas will disappear in 95 years." Obviously, a filled basin has lost its capacity to provide natural wetland functions of value to society; however, less intuitive is the impact of altered hydrology and spates of sediment inputs on wetland functions. Often wetlands are highlighted as providing numerous functions and values, including improving water quality. A fundamental property of wetlands to improve water quality is that they filter and retain sediment, and through physical, chemical, and biological processes, they sequester and transform pollutants. However, there is a tradeoff between the importance of sediment removal as a water quality benefit and maintaining the topographic life of wetland basins. Obviously, wetlands play an important role in improving environmental quality, especially controlling the offsite impacts of agricultural runoff on rivers, lakes, and reservoirs. Nevertheless, wetlands should only be used to remove sediment and other agricultural pollutants after agricultural best management practices have been implemented (Kuenzler 1990).

References

Adomaitis, V.A., H.A. Kantrud, and J.A. Shoesmith. 1967. Some chemical characteristics of aeolian deposits of snow-soil on prairie wetlands. Proc. North Dakota Acad. Sci. 21:65–69.

Adamus, P.R., and K. Brandt. 1990. Impacts on quality of inland wetlands of the United States: A survey of indicators, techniques, and applications of community-level biomonitoring data. U.S.E.P.A., Washington, DC, EPA/600/3-90/073.

Adamus, P.R. 1996. Bioindicators for assessing ecological integrity of prairie wetlands. U.S.E.P.A., Natl. Health and Environ. Res. Lab., W. Ecol. Div., Corvallis, Oregon, EPA/600/R-96/082.

Bellrose, F.C., and L.G. Brown. 1941. The effect of fluctuating water levels on the muskrat population of the Illinois River Valley. J. Wildl. Mgt. 5:206–12

Boto, K.G., and W.H. Patrick, Jr. 1978. Role of wetlands in the removal of suspended sediments. *In* Wetland Functions and Values: The State of Our Understanding, P.E. Greeson, J.R. Clark, and J.E. Clark (eds.), Amer. Resourc. Assoc., Proc. Natl. Symp. Wetlands, Minneapolis, Minnesota, pp. 479–89.

Castelle, A.J., C. Conolly, M. Emers, E.D. Metz, S. Meyer, M. Witter, S. Mauermann, T. Erickson, and S.S. Cooke. 1992. Wetland buffers: use and effectiveness. Washington Dep. Ecol., Shorelands and Coastal Zone Mgt. Prog., Adolfson Assoc., Inc., Olympia, Washington, Pub. No. 92–10.

Dahl, T.E. 2000. Status and trends of wetlands in the conterminous United States 1986 to 1997. USDI Fish and Wildl. Serv., Washington, DC, 82 pp.

Dieter, C.D. 1991. Water turbidity in tilled and untilled prairie wetlands. J. Freshw. Ecol. 6:185–189.

Dryer, P., L. Brooks, and N. Dietz. 1996. 1995 report: vegetation and sedimentation monitoring of undisturbed, cropland and waterbank wetlands in Wells County, North Dakota. Bluestem, Inc., Bismarck, North Dakota.

Duvick, D.N., and T.J. Blasing. 1981. A dendtoclimatic reconstruction of annual precipitation amounts in Iowa since 1680. Water Resourc. Res. 17:1183–89.

Ellis, M.M. 1936. Erosion silt as a factor in aquatic environments. Ecology 17:29–42.

Euliss, N.H., Jr., and G. Grodhaus. 1987. Management of midges and other invertebrates for waterfowl wintering in California. California Fish and Game 73:238–43.

Euliss, N.H., Jr., and S.W. Harris. 1987. Feeding ecology of northern pintails and green-winged teal wintering in California. J. Wildl. Mgt. 51:724–32.

Euliss, N.H., Jr., and D.M. Mushet. 1996. Water-level fluctuations in wetlands as a function of landscape condition in the prairie pothole region. Wetlands 16:587–93.

Frankforter, J.D., and S.A. Frenzel. 1995. Wetland water quality: Effect of local land use in the Platte River Basin, Nebraska [abs.]: Bul. North American Benthological Soc., v. 12, no. 1, p. 161.

Freeland, J.A., and J.L. Richardson. 1996. Soils and sediments as indicators of agricultural impacts on northern prairie wetlands. *In* S.A. Peterson, L. Carpenter, G. Guntenspergen, and L.M. Cowardin (eds.), Pilot Test of Wetland Condition Indicators in the Prairie Pothole Region of the United States. EPA/620/R-97/002, USEPA, Washington, DC, pp. 119–144.

Gleason, R.A. 1996. Influence of agricultural practices on sedimentation rates, aquatic invertebrates, and bird-use in prairie wetlands. Master's thesis, Humboldt State Univ., Arcata, California.

Gleason, R.A. 2001. Invertebrate egg and plant seedbanks in natural, restored, and drained wetlands in the prairie pothole region and potential effects of sedimentation on recolonization of hydrophytes and aquatic invertebrates. Ph.D. dissertation, South Dakota State Univ., Brookings, South Dakota.

Gleason, Robert A., and Ned H. Euliss, Jr. 1998. Sedimentation of prairie wetlands. Great Plains Research 8(1):97–112.

Goldsborough, L.G., and W.G. Crumpton. 1998. Distribution and environmental fate of pesticides in prairie wetlands. Great Plains Research 8:73–95.

Grue, C.E., L.R. DeWeese, P. Mineau, G.A. Swanson, J.R. Foster, P.M. Arnold, J. N. Huckins, P.J. Sheehan, W.K. Marshall, and A.P. Ludden. 1986. Potential impacts of agricultural chemicals on waterfowl and other wildlife inhabiting prairie wetlands: An evaluation of research needs and approaches. Trans. North American Wildl. and Natl. Resourc. Conf. 51:357–83.

Grue, C.E., M.W. Tome, T.A. Messmer, D.B. Henry, G.A. Swanson, and L.R. DeWeese. 1989. Agricultural chemicals and prairie pothole wetlands: Meeting the needs of the resource and the farmer-U.S. perspective. Trans. North American Wildl. and Natl. Resourc. Conf. 54:43–58.

Hammer, D.A. 1992. Designing constructed wetland systems to treat agricultural nonpoint source pollution. Ecol. Eng. 1:49–82.

Hartleb, C.F., J.D. Madsen, and C.W. Boylen. 1993. Environmental factors affecting seed germination in *Myriophyllum spicatum* L. Aquatic Bot. 45:15–25.

Jurik, T.W., Shih-Chin Wang, and A.G. van der Valk. 1994. Effects of sediment load on seedling emergence from wetland seedbanks. Wetlands, Vol. 14, No. 3, September 1994, pp. 159–165.

Kantrud, H.A., G.L. Krapu, and G.A. Swanson. 1989. Prairie basin wetlands of the Dakotas: a community profile. U.S. Fish and Wildl. Serv., Washington DC, Biol. Rep. 85.

Karl, T.R., and W.E. Riebsame. 1984. The identification of 10 to 20 year temperature and precipitation fluctuations in the contiguous United States. J. Climate and Applied Meteor. 23:950–66.

Krapu, G.L. 1974. Feeding ecology of pintail hens during reproduction. Auk 91:278–90.

Kuenzler, E.J. 1990. Wetlands as sediment and nutrient traps for lakes. *In* Enhancing the States' Lake and Wetland Management Programs, J. Taggart (ed.), Chicago: U.S.E.P.A., N. Amer. Lake Mgt. Soc., NE Illinois Planning Comm., Proc. Natl. Conf., pp. 105-12.

Lee, L.C., M.M. Brinson, W.J. Kleindl, P.M. Whited, M. Gilbert, W.L. Nutter, M.C. Rains, D.F. Whigham, and D. DeWald. 1997. Operational draft guidebook for the hydrogeomorphic assessment of temporary and seasonal prairie pothole wetlands. Seattle, Washington, 116+ pp.

Linz, G.M., R.A. Dolbeer, J.J. Hanzel, and L.E. Huffman. 1996. Controlling blackbird damage to sunflower and grain crops in the northern Great Plains. USDA, Animal and Plant Health Inspection Serv., Agric. Inf. Bul. No. 679, Washington, DC.

Long, C. 1991. National policy perspectives and issues regarding the prevention and control of nonpoint source pollution. USEPA-ORD Workshop on Nonpoint Source Pollution Control, Washington, DC.

Ludden, A.P., D.L. Frink, and D.H. Johnson. 1983. Water storage capacity of natural wetland depressions in the Devils Lake Basin of North Dakota. J. Soil and Water Conserv. 38:45–48.

Luo, H.R., L.M. Smith, B.L. Allen, and D.A. Haukos. 1997. Effects of sedimentation on playa wetland volume. Ecol. Appl., Vol. 7, No. 1, pp. 247–252.

Martin, D.B., and W.A. Hartman. 1987. The effect of cultivation on sediment composition and deposition in prairie potholes. Water, Air, and Soil Pollution 34:45–53.

Miller, M.R., and T.D. Nudds. 1996. Prairie landscape change and flooding in the Mississippi River Valley. Conserv. Biol. 10:847–53.

Mitsch, W.J., and J.G. Gosselink. 1993. Wetlands, 2nd ed., New York: Van Nostrand Reinhold.

Neely, R.K., and J. Baker. 1989. Nitrogen and phosphorus dynamics and the fate of agricultural runoff. *In* A.G. van der Valk (ed.), Northern Prairie Wetlands. Iowa State Univ. Press, Ames, pp. 92–131.

Neill, C., and J.C. Cornwell. 1992. Stable carbon, nitrogen, and sulfur isotopes in a prairie marsh food web. Wetlands 12:217–24.

Reeder, W.G. 1951. Stomach analysis of a group of shorebirds. Condor 53:43–45.

Renard, K.G., G.R. Foster, G.A. Weesies, D.K. McColl, and D.C. Yoder, coordinators. 1997. Predicting soil erosion by water: a guide to conservation planning with the revised universal soil loss equation (RUSLE). USDA, Agric. Handb. No. 703.

Richardson, J.L., J.L. Arndt, and J. Freeland. 1994. Wetland soils of the prairie pothole. Adv. in Agron. 52:121–71.

Rybicki, N.B., and V. Carter. 1986. Effect of sediment depth and sediment type on the survival of *Vallisneria americana* Michx grown from tubers. Aquatic Bot. 24:233–40.

Swanson, G.A., and H.F. Duebbert. 1989. Wetland habitats of waterfowl in the prairie pothole region. *In* Northern Prairie Wetlands, A.G. van der Valk, (ed.), Iowa State Univ. Press, Ames, Iowa, pp. 228–67.

van der Kamp, G., W.J. Stolte, and R.G. Clark. 1999. Drying out of small prairie wetlands after conversion of their catchments from cultivation to permanent brome grass. Hydrol. Sci. J. 44:387–397.

van der Valk, A.G., and C.B. Davis. 1976. The seed banks of prairie glacial marshes. Canadian J. Bot. 54:1832–38.

van der Valk, A.G., and R.L. Pederson. 1989. Seed banks and the management and restoration of natural vegetation. *In* M.A. Leck, V.T. Parker, and R.L. Simpson, (eds.), Ecology of Soil Seed Banks, Acad. Press, New York, New York, pp. 329—346.

Wang, Shih-Chin, T.W. Jurik, and A.G. van der Valk. 1994. Effects of sediment load on various stages in the life and death of cattail (*Typha* x *Glauca*). Wetlands 14:166–73.

Wayland, R. 1993. What progress in improving water quality? J. Soil and Water Conserv. 48:261–266.

Weinhold, C.E., and A.G. van der Valk. 1988. The impact of duration of drainage on the seedbanks of northern prairie wetlands. Can. J. Bot. 67:1878–1884.

IV.C Managing rice fields for wildlife and water quality (winter flooding of agricultural lands)

(Paul Rodrigue, NRCS Wetland Science Institute, Oxford, Mississippi, December 2001)

Purpose

This paper provides information on the benefits to both landowners and wildlife of managing rice fields for wildlife, principally post-harvest flooding. Readers are referred to a report detailing these benefits.

Contents

Rice fields (and other agricultural commodity fields) can be flooded after harvest to provide food and resting areas for migratory waterfowl. Landowners and wildlife both benefit from managing rice fields with post-harvest flooding.

Landowner benefits:
- Reduction of winter weeds resulting in reduced spring inputs (field preparation, herbicides)
- Reduction of red rice seeds (winter waterfowl can reduce red rice quantities in fields)
- Breakdown of crop residue
- Improved quality of discharge water (reduced sediment and nutrients)
- Opportunity for private recreation (hunting) or income from hunting leases

Waterfowl benefits:
- Food source (flooded residual grain, macroinvertebrates)
- Resting areas
- Limited shelter (crop stubble)

Operation

After the crop is harvested, the field is flooded for a portion of the waterfowl migratory period (typically November through February in the Lower Mississippi Valley). In some cases, particularly dry years, landowners may choose to flood the fields with pumped water. Some cost-share programs are available to offset costs in dry years. Some fields are disked or rolled before flooding.

The field should be flooded in stages to provide a new food source during the flood period. This can be accomplished by raising the outlet crest incrementally (e.g., installing only one or two stoplogs at a time) until the maximum storage is obtained.

Rice levees in the upper part of the field can be rebuilt to allow flooding of that portion of the field not controlled by the outlet structure.

Attachment

A copy of the Forest and Wildlife Research Center's Research *Advance* "Winter-Flooded Rice Fields Provide Waterfowl Habitat and Agricultural Values" is provided with this section for your information. It is also available at **<http://www.cfr.msstate.edu/fwrc/ricefields.pdf>**.

WINTER-FLOODED RICE FIELDS PROVIDE WATERFOWL HABITAT AND AGRICULTURAL VALUES

Gary Kramer

INTRODUCTION

Over 3 million acres of rice are grown annually in the United States, primarily in the Lower Mississippi Alluvial Valley (the Delta). In fact, the Delta regions of Arkansas, Louisiana, Mississippi and Missouri produce more than 2 million acres of rice annually. Americans truly enjoy rice as a side-dish, as evidenced by consumption of almost 27 pounds per person in 1998. Not only are people partial to rice, but it is also an important food for waterfowl, especially mallard, northern pintail, teal, and several species of geese.

Indeed. rice fields provide critical habitat for large numbers of North America's wintering waterfowl, shorebirds, and other wetland birds. However, only about 10% of the rice acreage in the Delta is currently managed to provide winter wetlands for waterfowl. Thus, extraordinary potential exists on rice lands for increasing the availability of wetland habitat for waterfowl and other waterbirds.

Scientists in the Forest and Wildlife Research Center (FWRC) at Mississippi State University recently investigated the potential values of winter-flooding rice fields and found the benefits were tremendous for both waterfowl and farmers. The team of FWRC researchers, composed of doctoral student Scott Manley, Dr. Rick Kaminski (Wildlife & Fisheries), Dr. Stephen Schoenholtz (Forestry), and research assistant Janet Dewey (Forestry), examined how different post-harvest treatments and winter-water management in ricefields affected soil erosion, water quality, rice-straw decomposition, weed control, and waterfowl food availability. The research was conducted during winters 1995-1997 and included 72 harvested rice fields, encompassing over 3,000 acres. Experiments were conducted in the major rice-producing areas in the Mississippi Delta, including Bolivar, Leflore, Sunflower, and Washington counties.

The scientists' primary objective was to test if winter-water management would benefit the environment, agriculture, and waterfowl. Another objective was to estimate potential cost savings in spring-field preparation to farmers who held water on rice fields during winter.

RESEARCH ADVANCES

Forest & Wildlife Research Center

B E N E F I T S

While environmental and wildlife conservation are truly important, practices which also decrease farming costs are most readily adopted by producers. Winter-water management of rice lands is such a practice.

Soil conservation and water-quality management in winter-flooded ricefields

Conserving soil and improving water quality are important in protecting our nation's natural resources. Experiments by FWRC scientists showed that winter flooding conserved soil and increased quality of runoff waters, especially when rice fields were not disked after harvest. Fall-disked fields allowed to drain freely after winter rains lost about 1,000 pounds of soil per acre (Figure 1). Fields with drain pipes closed to impound water during winter and with stubble left undisturbed after harvest lost only 31 pounds of soil per acre. Flooding rice fields not only reduces the impact of rain on exposed soils but also allows fields to act as settling basins and retain sediment and nutrients.

Winter-water management as a tool for spring-field preparation

By early spring, rice farmers must contend with challenges in field preparation for planting, such as disposal of remaining rice straw and growth of cool-season grasses and weeds. Reduction of rice straw is particularly challenging as it is resistant to physical degradation and decay, but it must be disposed of to facilitate planting. FWRC researchers found that winter flooding was as effective as fall disking in reducing by 53% the estimated 4.5 tons per acre of rice straw left after harvest (Figure 2). Elimination of fall-disking operations could save rice growers an average $14.13 per acre. The combination of fall disking and winter flooding reduced straw most significantly (68%), although disking incurs an added expense. The researchers also found that winter flooding inhibited germination and growth of cool-season grasses and weeds (Figure 3). If rice growers could eliminate aerial applications of spring "burn down" herbicides as a result of winter flooding rice fields, they could save an average of $13.19 per acre.

Winter-managed rice fields provide habitat for wetland wildlife

Researchers and rice growers know that rice left after harvest is an excellent source of food for waterfowl. However, the availability of 'waste rice' decreased 79-99% between harvest in August-September and early December when waterfowl typically arrive in the Delta in significant numbers. Researchers speculate that this decrease in waste rice during fall is due to a combination of factors, including germination of seed laying on the ground, decomposition, and consumption by rodents and birds. The decrease in waste grain has potentially serious implications for the foraging carrying capacity of rice fields and habitat needs for wintering waterfowl. Although availability of waste rice is much less than anticipated, the researchers found that flooded fields support winter populations of aquatic invertebrates, which are an important source of protein and minerals for waterfowl and shorebirds. Nevertheless, the researchers are concerned that winter food for ducks and geese may be limited in Delta rice fields; thus, a new research initiative is underway to validate these findings and devise management strategies to counter possible food shortages for wintering waterfowl.

P R O C E D U R E S

Managing winter water in rice fields is relatively easy and inexpensive for rice growers because rice is grown in an aquatic setting. By following the procedures below, a winter flooding project should be successful.

1. Maintain water control systems and levees used for rice culture to impound winter rainfall. These sites are ideal for developing wintering habitat.

2. Consider refraining from fall disking to save money and prevent incorporation of 'red rice' seeds into soils.

3. Hold water on fields throughout winter.

4. As spring approaches, drain fields gradually to concentrate aquatic invertebrates and expose mud-flats for feeding waterfowl and shorebirds.

Overall, winter flooding of harvested rice fields was determined to be a valuable conservation practice that benefits the environment, farm operations, and waterfowl. Winter-water management is an excellent example of how agriculture can be compatible with wildlife management. This research also reaffirmed the importance of conserving other natural habitats, such as bottomland hardwood forests and moist-soil wetlands, to provide alternative foraging areas for wintering waterfowl when waste rice is in short supply.

Figure 1

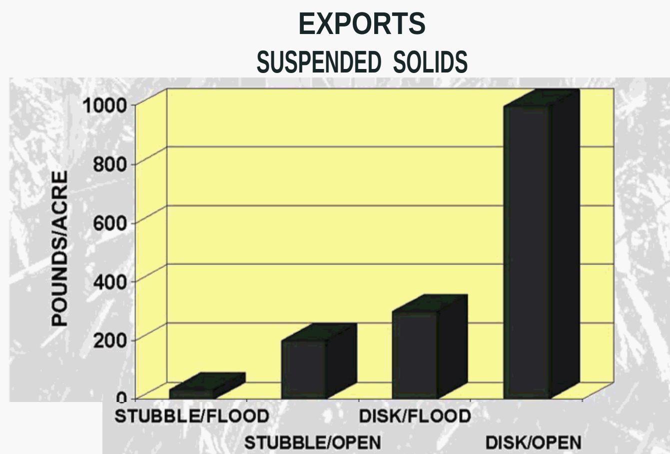


Figure 2

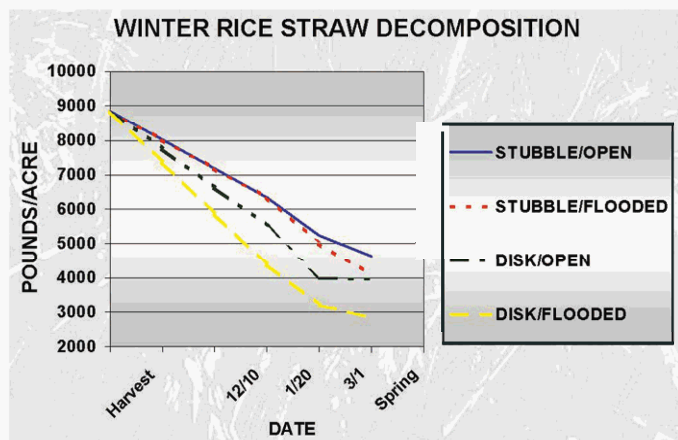
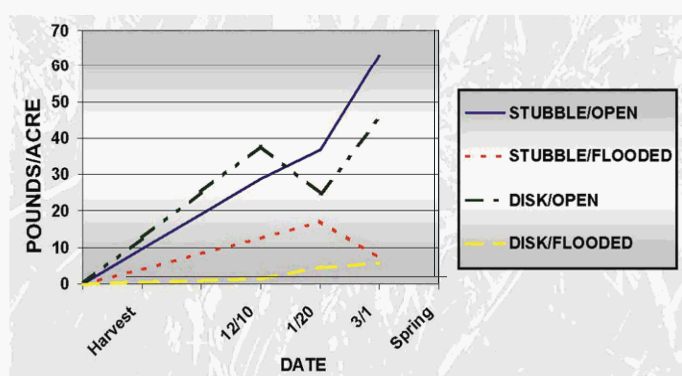


Figure 3

WINTER WEED GROWTH IN RICEFIELDS



RESEARCH ADVANCES
December 1999 Vol. 4, No. 1

Research conducted by
Richard Kaminski
Scott Manley
Stephen Schoenholtz
Janet Dewey

For more information contact:
Bob L. Karr,
Interim Director
Forest & Wildlife
Research Center
Box 9680
Mississippi State, MS 39762

662-325-2952
Fax: 662-325-8726
bkarr@cfr.msstate.edu
http://www.cfr.msstate.edu

Karen Brasher. Editor

FWRC-WF-148

ACKNOWLEDGEMENTS

Financial sponsors of this research were Dow AgriScience. Inc.; the Institute for and and Waterfowl Research, Ducks Unlimited, Inc.; Federal Aid in Wildlife oration through the Mississippi Department of Wildlife, Fisheries and Mississippi Rice Promotion Board; Mississippi Water Resources Research Institute; National Fish and Wildlife Foundation; U.S. Geological Service's Patuxent Wildlife Research Center; U.S. and Wildlife Service's Lower Mississippi Valley Joint Venture; and Mississippi State University's Forest and Wildlife Research Center. Field research was conducted on the following farms Aguzzi, Arant, Circle-H, Morgan, Murrell, Opossum Ridge, and Tackette. Technical and logistical assistance was provided by Delta Wildlife Foundation, Mississippi State University Extension Service, Mississippi Department of Agriculture and Commerce, Mississippi Farm Bureau Federation, U.S. Department of Agriculture's Natural Resources Conservation Service, and the U.S. Fish and Wildlife Service's Mississippi Wetland Management District and the Noxubee National Wildlife Refuge.

This Research *Advance* was based upon the following doctoral dissertation: Manley, S.W. 1999. Ecological and agricultural values of winter-flooded ricefields in Mississippi. Ph.D. Dissertation, Department of Wildlife and Fisheries, Mississippi State University.

IV.D Wetland management options in the Lower Mississippi River Alluvial Valley

(Phil Covington, regional biologist, Ducks Unlimited, Inc., North Little Rock, Arkansas, December 2001)

Purpose

This paper presents wetland management practices for habitats that have a proven track record of increasing habitat value of wetlands to wildlife in the Lower Mississippi River Alluvial Valley (LMRAV). Characterization and management considerations for wetland habitats and associated wildlife areas occurring in the LMRAV include bottomland hardwoods, moist-soil wetlands, emergent marshes, shrub-scrub swamps, food plots, and fishless ponds.

Contents

The Lower Mississippi River Alluvial Valley has lost more than 70 percent of its bottomland hardwood forested wetlands over the past 200 years. Of the original 24.7 million acres of bottomland hardwood forests, only about 7 million acres remain. Much of this loss occurred in the 1960's when bottomland forests were cleared for soybean production. During the 1950s to 1970s, annual losses exceeded 290,000 acres. Efforts by private organizations as well as programs of local, State, and Federal Governments are restoring some of the wetlands lost to the region.

One of these efforts that has been highly successful is the Wetland Reserve Program of the U.S. Department of Agriculture, Natural Resources Conservation Service. This program restores wetlands by purchasing easements from willing landowners. Marginal cropland is taken out of production, and vegetation and water are restored for wetland benefits. Obviously this program, or any other, will never replace all the wetlands that historically occurred, but to date (July 2000) the Wetland Reserve Program has enrolled 312,090 acres in Arkansas, Louisiana, and Mississippi. With proper management each restored acre could contribute significantly to the reestablishment of wetlands throughout the Mississippi Valley.

A Wetland Plan of Operations is developed as part of the restoration and enhancement activity. This plan outlines objectives that match the wetland functions that the landowner values most. In many cases this means attracting waterfowl; however, some landowners place high value on shorebirds, wading birds, or frogs and salamanders. It is important to know what is being managed for, as no wetland can supply all needs for all wetland wildlife species during all life phases.

In many instances trees have been planted, wetlands replaced, and water control structures put in place. The next step is to begin to understand the fundamental principles behind established wetland management strategies.

The golden rule among physicians is "first do no harm." This should also be the guiding thought among wetland managers. Our statement of purpose should be "emulate Mother Nature and never flood the marsh to the same depth and duration in successive years and neither flood nor drain on the same calendar date year after year." Remember the letters DDT, they stand for **depth, duration,** and **timing**. To "do no harm" to wetland plants, particularly trees, the DDT must be varied among and within years.

A wetland system provides opportunity to manage several if not all of the following wetland habitats: bottomland hardwoods, moist-soil wetlands, emergent marshes, shrub/scrub swamps, food plots, and fishless ponds. Each habitat type is described individually.

Bottomland hardwoods

Bottomland hardwood wetlands are forested wetlands comprised of trees, shrubs, forbs, and grasses that can withstand flooding at various depths, duration, and timing. Susceptibility to flooding was considered when tree species were selected for the site in the restoration plan. Those species that can withstand longer periods of inundation during the growing season are planted on the lower sites. Typically, if a site exhibits a wide range of elevation, cypress and tupelo are planted at the lowest site. Green ash, overcup oak, and nuttall oak may occupy the next elevational shelf, followed by water and willow oak, and possibly shumard and cherrybark oak at the highest elevations.

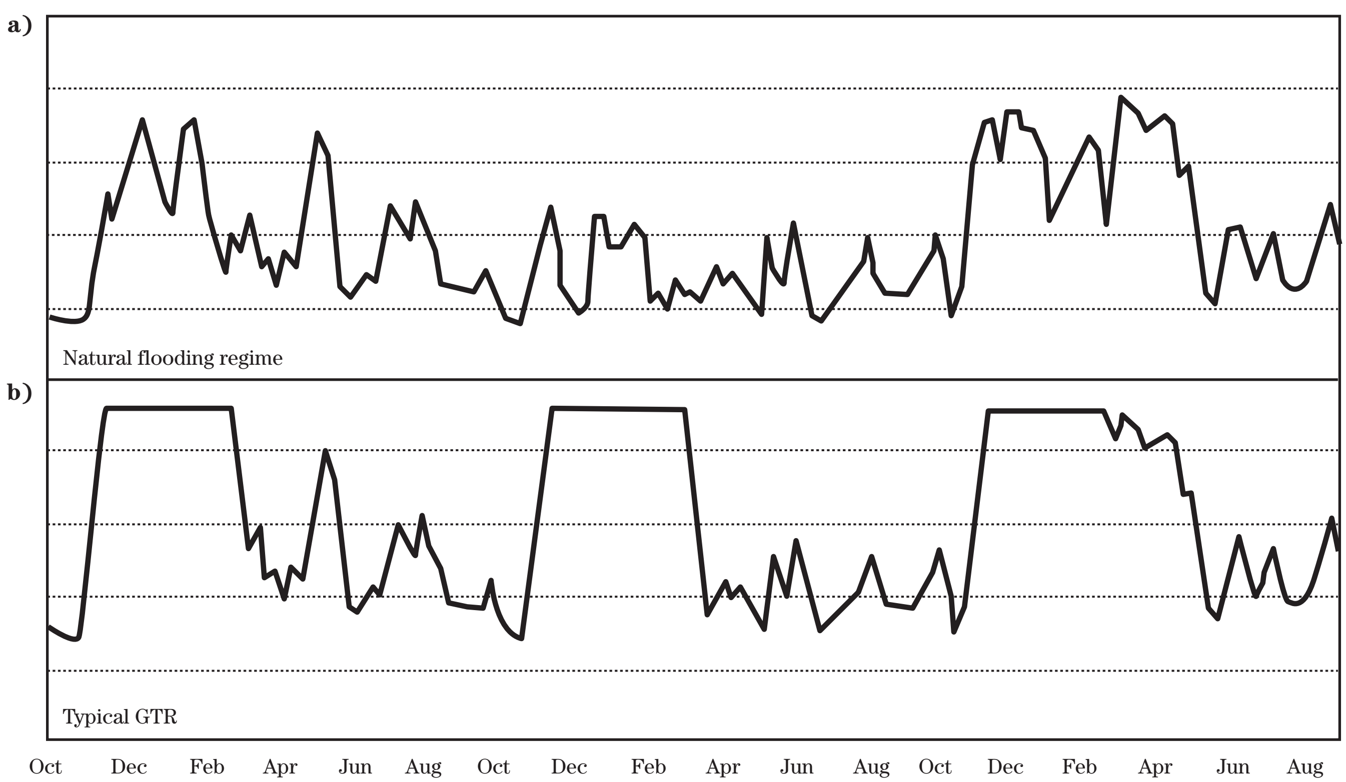
Species that are extremely tolerant to flooding, such as cypress, still do well on elevated sites; however, those species intolerant to prolonged flooding, such as cherrybark, will not do well at lower elevation.

If the bottomland hardwood site is surrounded by a levee and water levels are controlled, the site being managed is a greentree reservoir. In addition to the DDT rule, it is extremely important that flood sensitive trees **not** be flooded each year during the growing season. Bottomland trees can handle flooding during dormancy, but if roots are under water during the growing season, less tolerant trees (cherrybark, shumard, and cow oak) will be stressed. If this occurs frequently or for prolonged periods during the growing season, the oaks will most likely die. Avoid annual flooding before December and drain flood sensitive trees by March 15. Some evidence has shown that early fall flooding is more detrimental than late spring flooding. As mentioned, avoid flooding and draining on the same date each year. Varying the depth of flooding during a season is important (fig. IV.D–1). Several years of improper flooding result in tree stress exemplified by yellowish leaves, dead branches, acorn crop failure, and swollen and cracked tissue at water level. Improper flooding over decades results in dead hardwoods and an increasing number of flood-tolerant trees, such as cypress, tupelo, and overcup oak.

Moist-soil wetlands

Managing for moist-soil plants is accomplished by the periodic introduction and removal of water on a wetland to encourage seed-producing annuals to flourish while discouraging nontarget plants, such as coffee bean and cocklebur. Planting seeds is not necessary because seeds of native plants are abundant in frequently flooded soils. Wild millet, sprangletop, and smartweed are examples of desirable moist-soil plants, which provide nutrients to seed-eating ducks, such as mallards, green-winged teal, northern pintail, and ringnecks. Slow drawdowns during the growing season should result in a wide range of appropriate plants

Figure IV.D–1 Comparison of a natural flooding regime to that of a typical GTR over a 3-year period (a) and within the same year (b) (note the variability both among years (a) and within years (b))



germinating on exposed mud flats. An additional benefit to a slow (1 to 2 inches a day) drawdown is the prolonged concentration of bugs, crayfish, frogs, and small fish, which in turn attract foraging ducks, herons, egrets, bitterns, and shorebirds. Drawdowns in March and April generally result in smartweed and wild millet; May/June drawdowns encourage wild millet and beggarticks, while later season drawdowns favor sprangletop and panicgrass. Late, rapid drawdowns may result in undesirable species, such as cocklebur or coffee bean. It is not uncommon to produce 2,000 pounds per acre of seed in a properly managed moist-soil marsh. Over a period natural plant succession will favor perennial plants over annuals. Perennials do not produce as much seed as annuals; therefore, if seed production is the goal, disking, burning, or mowing the marsh is necessary every 3 or 4 years to set plant succession back to the annual stage.

Flooding of the moist-soil vegetation should be timed to coincide with the arrival of the target species (table IV.D.–1). Seed decomposition should also be considered. After 90 days of flooding, wild millet seed is 57 percent decomposed, while smartweed seed has decomposed by only 21 percent (table IV.D–2). Generally, seeds with hard seed coats like smartweeds decompose more slowly than soft-shelled seeds. Sora rail and teal use flooded moist-soil August through October. Managers interested in waterfowl hunting may prefer to start flooding a few weeks before waterfowl season. If more than one moist-soil wetland is available, stagger draining and flooding over the period. This adds to plant diversity and prolongs habitat availability. Studies in Missouri found that of the 156 bird species that use moist-soil wetlands, 131 prefer water depths of 10 inches or less (fig. IV.D–2). A dabbling duck that has to tip up to feed on submerged plant roots is spending additional energy.

Figure IV.D–2 Preferred water depths for wetland birds commonly associated with moist-soil habitats

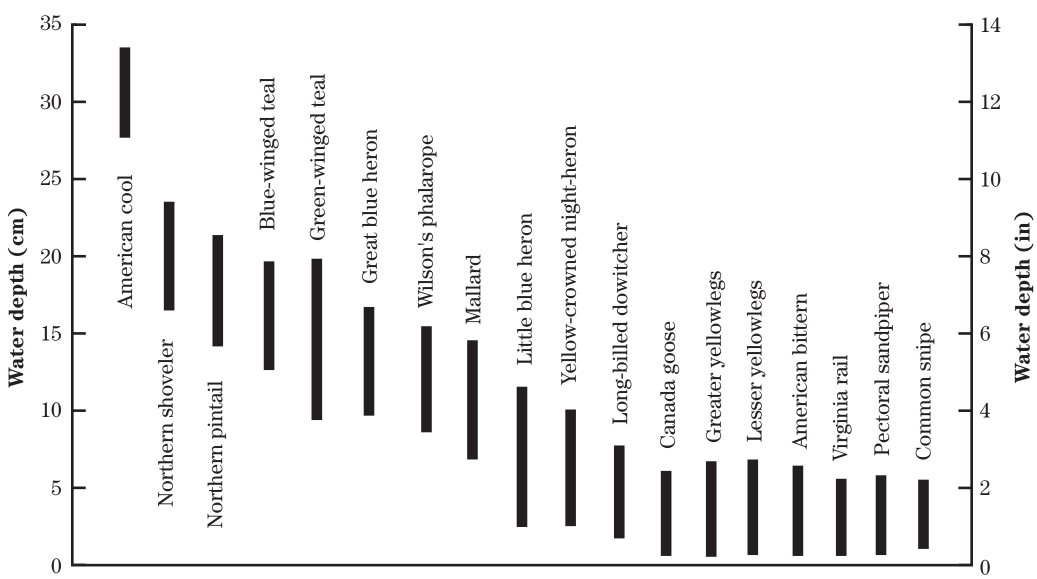


Table IV.D–1 Water level scenarios for target species on three moist-soil impoundments and associated waterbird response

Period	Unit A — Water level scenario	Unit A — Water level response	Unit B — Water level scenario	Unit B — Water level response	Unit C — Water level scenario	Unit C — Water level response
Early fall	Dry	None	Dry	None	Gradual flooding starting 15 days before the peak of early fall migrants; water depth never over 4 inches.	Good use immediately; high use by teal, pintails, and rails within 2 weeks.
Mid fall	Dry	None	Flood in weekly 1- to 2-inch increments over a 4-week period.	Excellent use by pintails, gadwalls, and wigeons.	Continued flooding through September.	Excellent use by rails and waterfowl.
Late fall	Flood in weekly 2- to 4-inch increments over a 4- to 6-week period.	Excellent use immediately by mallards and Canada geese.	Continues flooding, but not to full functional capacity.	Excellent use by mallards and Canada geese.	Continued flooding to full functional capacity.	Good use by mallards and Canada geese.
Winter	Maintain flooding below full functional capacity.	Good use by mallards and Canada geese when water is ice-free.	Maintain flooding below full function capacity.	Good use by mallards and Canada geese when water is ice-free.	Continued flooding to full pool.	Good use by mallards and Canada geese when water is ice-free.
Late winter	Schedule slow drawdown to match northward movement of migrant waterfowl.	Excellent use by mallards, pintails, wigeons, and Canada geese.	Schedule slow drawdown to match northward movement of early migrating waterfowl.	Excellent use by mallards, pintails, wigeons, and Canada geese.	Schedule slow drawdown to match northward movement of waterfowl.	Good use by mallards and Canada geese when water is ice-free.
Early spring	Continued slow drawdown to be completed by 1 May.	Excellent use by teals, shovelers, shorebirds, and herons.	Drawdown completed by 15 April.	Excellent shorebird use.	Drawdown completed by 15 April.	Excellent shorebird use.

Table IV.D–2 Deterioration of selected seeds after 90 days of flooding

Plant name	Decomposition (%)
Soybean	86
Barnyardgrass	57
Corn	50
Common buckwheat	45
Milo	42
Giant bristlegrass	22
Pennsylvania smartweed	21
Cultivated rice	19
Water pak (acorns)	4
Hemp sesbania	4
Horned beakrush	2
Saltmarsh bulrush	1

Emergent marshes

Emergent marshes generally are 12 to 30 inches deep and contain vegetation that is rooted in the soil and emerges above the water surface. Typical emergent plants include cattail, bulrush, and rice cutgrass. These sites are valuable nesting and brood rearing habitat for wading birds, such as rail, bittern, grebes, and coots. Emergent marshes are also used by winter migrant waterbirds for feeding, roosting, and resting. Maximum use by most bird species is realized when emergent plants cover 50 percent of the water surface. This open water/emergent cover ratio is called a hemimarsh. Allowing natural succession is important because some species, such as king rail, prefer less than 50 percent emergent cover for foraging while others like the least bittern often nest in marshes with more than 50 percent emergent cover. Emergent marsh succession can be economically interrupted when shrub and tree stems reach 2 inches in diameter. At this stage a bushhog mower and a heavy disc can be used to set succession back to the annual grass stage. Grebes, coots, and American bitterns use floating nests that are attached to emergent vegetation, so any extreme water level fluctuation during nesting could cause egg drowning or nest tipping. Another management practice involves draining emergent marshes after nesting birds have fledged. This attracts fall migrating shorebirds, most of which prefer feeding in 1 to 2 inches of relatively open water (less than 33% vegetative cover) that is slowly drained, thereby, concentrating bugs and small fishes in ever decreasing shallow depressions.

Shrub/scrub swamps

Shrub/scrub swamps are typified by willow wetlands with interspersions of other soft wood species, such as buttonbush, and perennial marsh vegetation. In some areas shrub/scrub sites are transitional between emergent marshes and forested wetlands. Historically, wetland managers did not fully understand the contribution of shrub/scrub habitat to a healthy wetland system. Decaying willow leaves provide nutrients for many aquatic bugs that in turn provide protein for foraging water birds, fish, amphibians, and other wetland inhabitants. Studies have documented over 25 pounds of bugs produced in an acre of flooded willows. Buttonbush seeds also provide important nutrients, such as fatty acids. Wood ducks and wintering gadwalls are especially attracted to this wetland type. However, the primary contribution of shrub/scrub habitat usually is not food production. It has been documented that on a cold night a mallard that roosts in a flooded corn field instead of the warmer confines of a willow or buttonbush wetland must consume 42 kernels of corn or 3,500 sowbugs the following day to offset the energy spent in maintaining adequate body heat throughout the previous night. The same mallard could have avoided burning that energy if it had roosted in a shrub/scrub wetland.

Food plots

In the context of wetland management, food plots are generally employed to attract waterfowl. Food plots are more beneficial if moist-soil weeds are interspersed with the planted crop. This interspersion ensures nutritional variety and an increase in wetland bugs if the use of pesticide is avoided. Corn and rice are high in carbohydrates and are readily available packets of concentrated energy that help ducks maintain body temperature throughout winter. Historically, acorns, which are equally high in carbohydrates, met this requirement. On a regional basis food plots probably are not as important as one might think since Arkansas has many acres of flooded rice and other harvested grains during the winter, and flooded acorns are still present, but in reduced quantities. Food plots are, however, important tools when maintaining an early successional stage of wetland progression. When choosing the food plot mix, soybeans should be avoided because they are relatively low in carbohydrates and waterfowl are unable to digest the protein. When consumed in dry form, soybeans can swell causing severe problems to waterfowl.

Fishless ponds

Fishless ponds are depressional wetlands at high elevations that escape seasonal flooding and are subject to frequent dry periods. As a result, no fish or bullfrog tadpoles are present to feed on the resident amphibians' eggs or young. The northern spring peeper, northern crawfish frog, and eastern spade foot are some of the frogs found in these ponds. Fishless wooded ponds often have marbled, spotted, and ringed salamanders. The central newt is seldom numerous in ponds that contain fish. Local resident species have evolved special techniques to cope with periodic drying of the pond. Some simply migrate to nearby wetlands while others burrow underground and await the return of water. Most can complete metamorphosis in less than 3 months. Frogs and salamanders are important links in the food chain, and they contribute to a healthy wetland system.

Nutritional requirements

Birds require different foods at different times of the year depending on their current physiological needs. For instance, when a female mallard first arrives in southern areas in early fall, she requires a wide diversity of nutrients found in seeds, stems, and rootlets of various moist-soil plants. She also feeds on bugs in moist-soil wetlands to acquire the protein necessary to replace feathers during the fall molt. As autumn progresses she needs high-energy fat and carbohydrates to maintain body temperature. She also acquires fat for pairing needs and to sustain local movements. For this, she feeds on rice, corn, soybeans, and acorns and seeds in flooded timber. During late winter she prepares her body for another feather molt (protein) and eventual egg laying (calcium and protein). Meanwhile, she begins storing fat (fatty acids) around her gut to provide energy for the flight north to the breeding grounds. The best place to meet these nutritional requirements is in shallowly flooded bottomland hardwoods dominated by red oaks. Bugs and worms provide protein for feather molt and egg albumin production, while fingernail clams provide calcium for the eggshell. Acorns are high in fatty acids and provide much of the stored energy necessary for the northward migration. Good conditions in the southern areas allow the mallard to meet nutritional needs, and she can immediately commence laying eggs upon arrival at the northern nesting grounds.

Regional wetland complexes

A marsh is an important component of all the neighboring marshes that comprise the county, the State of Arkansas, the Mississippi Flyway, the four continental flyways, and ultimately the global migratory bird community. Local wetland creatures, such as turtles, crayfish, and muskrats, can often make a living within an individual marsh. Many migratory waterbirds, however, need marshes stretching from Canada to South America. A variety of wetland types located throughout the flyway ensures that each species can meet its physiological requirements during each stage of its life. Most WRP lands are not large enough to provide all of the previously mentioned wetland types. Studies indicate that if a mallard cannot find all the resources it needs for survival within a 12-mile radius, it will be forced to seek those resources elsewhere. Each 12-mile circle should contain bottomland forest, green tree reservoirs, shrub/scrub swamps, emergent marsh, moist soil wetlands, fishless ponds, and flooded cropland. Furthermore, each of these wetland types should occur in several progressional stages of succession. This means each individual marsh, neighboring marshes, and nearby State and Federally owned marshes all contribute to the wetland diversity that is necessary for a healthy environment.

To Contents

IV.E Woody plant species for restoration and enhancement in the South and Southeast

(Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001)

Purpose

This section provides information on selected woody plants that occur natively in the bottomlands and other wetland habitats in the south and southeast. These species are being used in wetland restoration and enhancement projects and are recommended for use. Each species sheet in this section has information on the plants general economic and wildlife use, identification characteristics, habitat and geographic distribution, establishment, and management techniques.

Contents

The species included in this section are shown in the table below.

IV.E.1 Red maple (*Acer rubrum* L.) (ACRU)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph, Norman Melvin; and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

Red maple is well adapted to occasionally flooded bottomland areas and upland areas with moist soils. It is used for furniture, turney, woodenware, fuel, veneer, crates, boxes, flooring, slack cooperage, interior finish, and novelties. Red maple is suited as a source of soft mast that grows in flooded areas. It is also valuable as source of nectar and pollen in the early spring stimulation of bees. Red maple is also sometimes used as an ornamental.

Characteristics

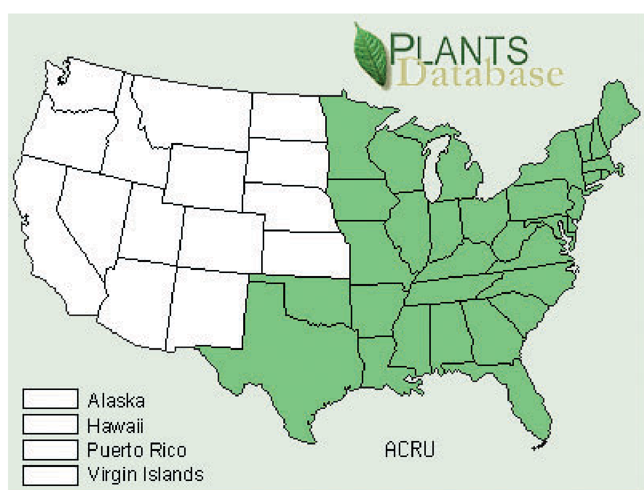
Red maple grows to a height of 100 feet and a diameter of 3 feet. The bark on younger trees is smooth and light gray. On large trunks, it is reddish brown and furrowed with thin scales. The leaves are opposite, three- to five-lobed, pointed, with a coarsely toothed margin. They are usually 2 to 6 inches in diameter. The leaves are light green above, pale below, and turn red in early autumn. The fruit is a pair of winged seeds 1 to 1.5 inches long borne on drooping stems 3 to 4 inches long. This species flowers in early spring before the leaves develop. Fruit matures in late April and May (depending upon its occurrence within its range).

Adaptability

Red maple occurs in a variety of habitats and on various soils. It is most frequent on well-drained, moist soils, but also occurs commonly on drier ridges and exposed slopes as well as swamps. In the Mississippi River Alluvial Valley, red maple is adapted to the Southern Mississippi Valley Alluvium, Southern Mississippi Valley Silty Uplands. It is tolerant of periodic flooding from January to June.

Establishment

Red maple is established by seedlings. Adequate size for planting is generally 1-year-old seedlings. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.



Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

Notes

Several wetland varieties of this species occur throughout the range. The Trident red maple (*A. rubrum* var. *trilobum*) and Drummond's red maple (*A. rubrum* var. *drummondii*) both occur on wetter sites than the typical red maple. Both of these two varieties have a wetland indicator status designation of FACW suggesting they have a greater frequency of occurrence in a wetland than the typical red maple. Be aware these varieties occur and use them to your advantage in a restoration effort.

IV.E.2 Sweet pecan (*Carya illinoensis* (Wangenh.) K. Koch) (CAIL2)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

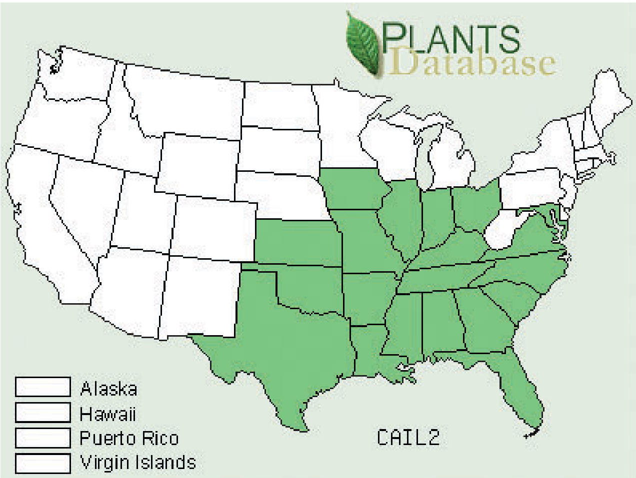
Sweet pecan is well adapted to occasionally flooded bottomland areas. It is used for flooring, furniture, veneer, and edible nuts. Sweet pecan is suited as a wildlife food source for birds and mammals. It is also an attractive shade tree.

Characteristics

Sweet pecan grows to a height of 130 feet and a diameter of 2 to 4 feet. The bark is gray, rough, hard, and tight. It is sometimes broken into scales. Bark on young branches is smooth, but it fractures with age. The leaves are alternate and compound with 9 to 17 leaflets. The entire leaf is 12 to 20 inches long with leaflets 4 to 8 inches long and 2 inches wide. The leaflets are toothed on the margins and long-pointed with the upper side wider than the lower, and appearing to curve downward. The fruit is a nut with a four-winged, pointed, thin husk. It is 1 to 2 inches long and 0.5 to 1 inch in diameter.

Adaptability

Sweet pecan occurs in the bottomlands of the Mississippi River Valley and the southeast coast from Iowa and Ohio southward to the gulf, into eastern Texas, and into central Mexico. It is adapted to the Southern Mississippi Valley Alluvium, Southern Mississippi Valley Silty Uplands, and the Arkansas Valley and Ridges, but can commonly occur on well-drained loam soils. It is tolerant of periodic flooding from January to April, but is intolerant of prolonged flooding and rarely occurs on clay flats. Sweet pecan is usually on moderately well drained alluvial loams of first bottoms.



Establishment

Sweet pecan is established by seedlings. Adequate size for planting is generally 1-year-old seedlings. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet. The least desirable trees should be removed first during a thinning; those poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

Notes

This species has been extensively cultivated for commercial nut production. Its current distribution as projected on the distribution map exceeds its native range; from southern Illinois and Indiana southward to the gulf, into eastern Texas, and sporadically into central Mexico. Using this species for restoration in a strictly purist sense should not go beyond this original range.

IV.E.3 Sugarberry (*Celtis laevigata* Willd.) (CELA)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

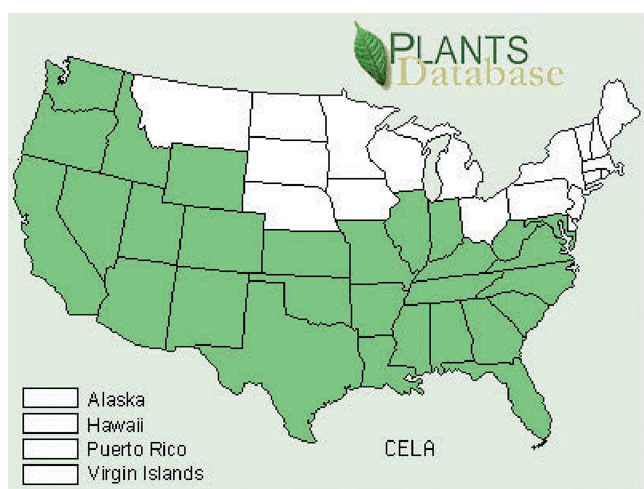
Sugarberry is well adapted to frequently flooded bottomland areas. It is used for boxes, baskets, crating, furniture, and burial boxes. Sugarberry is suited as a source of soft mast that grows in extensively flooded areas. It is also an attractive shade tree.

Characteristics

Sugarberry grows to a height of 80 feet on wet soils and 40 to 50 feet on uplands. It may attain a diameter of 2 to 4 feet. The bark is distinctively smooth and light gray, becoming spotted with age and developing prominent warty projections. The leaves are egg-shaped and taper-pointed. The margins are untoothed except for a few coarse teeth at the tip. They are usually 2 to 5 inches long and 0.75 inch to 1.5 inches wide. The fruit is a 0.25-inch diameter berry, which is at first orange red or yellow and later turns reddish brown. It has little flesh and a large pit.

Adaptability

Sugarberry commonly occurs on poorly drained, alluvial clays of broad flats and shallow sloughs in bottomland areas and along river flood plains throughout its range. It is widely distributed on bottomlands and occurs on moist upland sites. Within Lower Mississippi River Alluvial Valley, it is adapted to the Southern Mississippi Valley Alluvium, Southern Mississippi Valley Silty Uplands, and the Western Coastal Plain. It is tolerant of continuous flooding from January to May. Although the recent geographic distribution map identifies it as occurring widely, it is a major component of bottomland forests from southeastern Virginia to mid-Texas and Oklahoma with isolated collections elsewhere.



Establishment

Sugarberry is established by seedlings. The 1- or 2-year-old seedlings are generally adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where the canopy is less than 12 feet in height. Fire protection is necessary at all times.

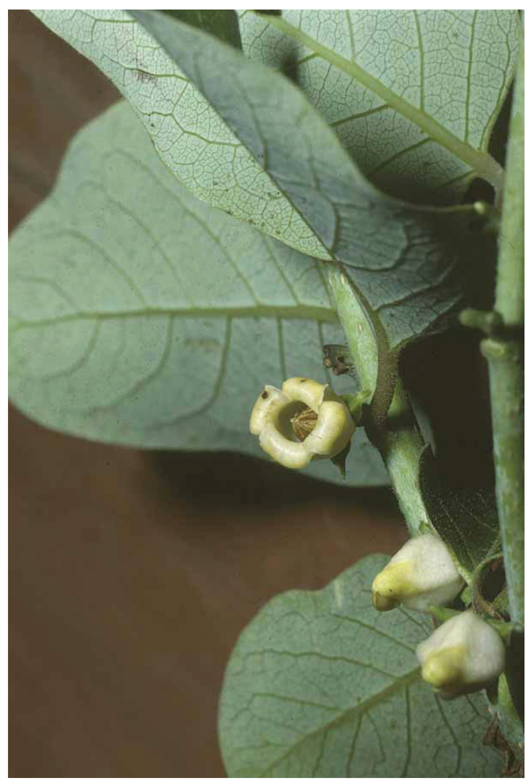
The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning; those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.4 Persimmon (*Diospyros virginiana* L.) (DIVI5)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)



General use

Persimmon is well adapted to frequently flooded bottomland areas and upland areas with moist soils. It is used for shuttles, golf clubs heads, shoelasts, billiard cues, brush backs, and novelties. Persimmon is suited as a source of soft mast that will grow in flooded areas. It is also valuable for honey production.

Characteristics

Persimmon grows to a height of 50 feet and a diameter of 18 inches. The bark on older trunks is almost black and is broken into nearly square blocks. The leaves are oval and sharp pointed with a smooth or wavy margin. They are usually 4 to 6 inches long. The leaves are shiny and dark green above and paler below. This species occurs in separate male and female trees. The male trees do not produce fruit, but do flower producing pollen and nectar. The fruit produced by the female trees is round, pulpy, orange to brown, and is 1.5 inches in diameter, astringent when unripe and sweet at maturity especially after frost. It contains three to eight large, hard, smooth, brown seeds 0.5 inch long.

Adaptability

Persimmon occurs on a variety of sites, but is most common on alluvial soils composed of clay and heavy loam. It also occurs on light, sandy, well-drained sites. In the Mississippi River Alluvial Valley, persimmon is adapted to the Southern Mississippi Valley Alluvium and Southern Mississippi Valley Silty Uplands. It is tolerant of continuous flooding from January to May.

Establishment

Persimmon is established by seedlings. The 1-year-old seedlings are generally adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates:	December 1 to March 31
Plant spacing:	12- by 12-foot (302 trees per acre)
Planting depth:	At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

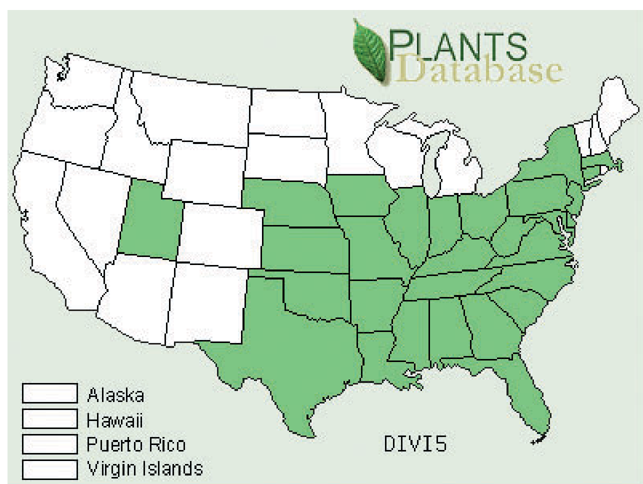
The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning; those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

Notes

The seeds of persimmon are dispersed by animals that eat the fruits and deposit them elsewhere in scat. For this reason it is commonly one of the first mast species to colonize the interior of restoration areas, not just the peripheral edges.

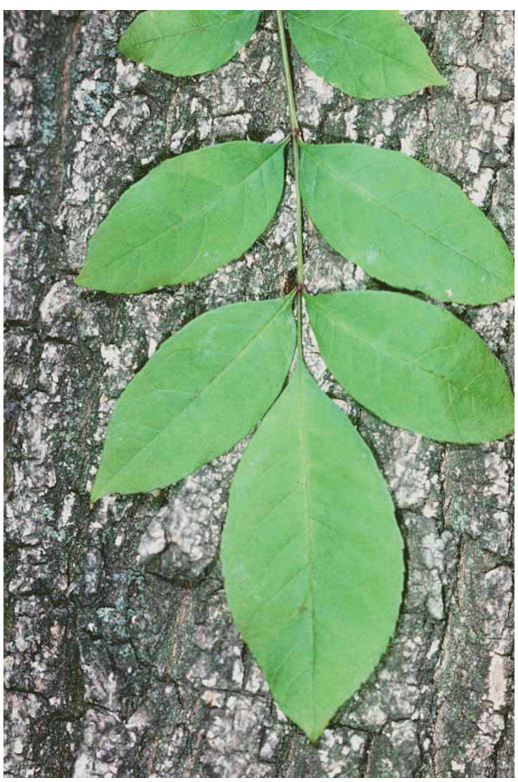


IV.E.5 Green ash (*Fraxinus pennsylvanica* Marsh.) (FRPE)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General Use

Green ash is well adapted to a variety of sites from frequently flooded bottomland areas to well-drained and droughty uplands. It is used for tool handles, baseball bats, boat oars, skis, agricultural implements, furniture, trunks, plywood, and cooperage. Green ash is suited as wildlife food source for small mammals and birds. It is also an attractive shade tree.



Characteristics

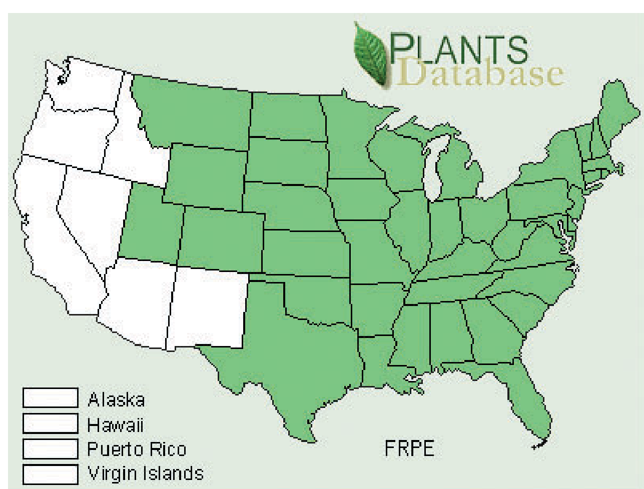
Green ash grows to a height of 40 to 60 feet and a diameter of 1 to 2 feet. It has slender, spreading branches forming a compact crown. The bark is tinged red with netted fissures and ridges. The 9- to 12-inch-long leaves are opposite and compound with 7 to 9 short-stalked leaflets. The leaflets are pointed on both ends and toothed along the margins. They are bright green on both sides. The fruit is a seed winged to more than half its length. It is 1 to 1.5 inches long.

Adaptability

Green ash is tolerant of continuous flooding from January to May. This tree generally is on poorly drained alluvial clays and clay loams. It may be planted on well-drained and droughty upland soils and has been used as a shelterbelt tree and mine reclamation plant.

Establishment

Green ash is established by seedlings. The 1-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has



stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

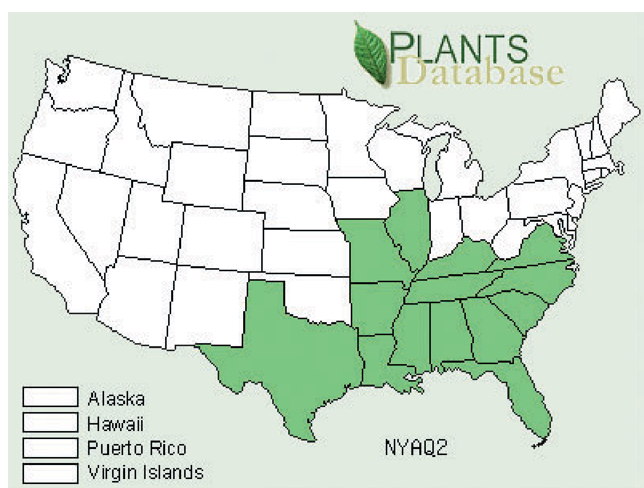
The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning; those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.6 Water tupelo (*Nyssa aquatica* L.) (NYAQ2)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)



General use

Water tupelo is well adapted to frequently flooded bottomland areas. It is used for woodenware, broom handles, fruit and vegetable packages, pulp, box boards, interior trim, and cigar boxes. Water tupelo is suited as a source of soft mast that will grow in extensively flooded areas. It is also valuable for honey production.

Characteristics

Water tupelo grows to a height of 100 feet and a diameter of 4 to 5 feet at the buttressed roots. The bark on large trunks is rather thick, dark brown or dark gray brown, and has deep longitudinal fissures roughened on the surface by small scales. The leaves are egg-shaped, sometimes long-pointed, and somewhat wedge-shaped at the base with a few coarse teeth on the margins. They are usually 5 to 9 inches long and 2 to 4 inches wide. The fruit is a purple, plum or olive-shaped berry about 1 inch long. It has a flattened stone inside and is borne on a slender stalk 3 to 4 inches long.

Adaptability

Water tupelo occurs in deep swamps, bottomlands, and along watercourses throughout the Coastal Plain of the South and Southeast extending into the Midwest States. It generally is on poorly drained alluvial clays to silt loams. Water tupelo is adapted to the Southern Mississippi Valley Alluvium and the Southern Mississippi Valley Silty Uplands. It is tolerant of continuous flooding from January to June.

Establishment

Water tupelo is established by seedlings. The 1- to 2-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

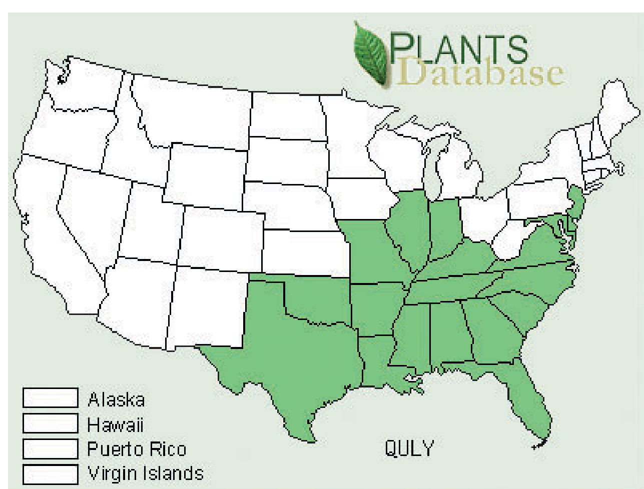
The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.7 Overcup oak (*Quercus lyrata* Walt.) (QULY)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph, Jody Pagan, NRCS, Arkansas; State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)



General use

Overcup oak is in the white oak group. It is well adapted to frequently flooded bottomland areas. It is used for flooring, furniture, construction, millwork, cooperage, crossties, handles, and fuel. Timber quality is not as high as nuttall oak. Overcup oak is an especially valuable wildlife food source because it is the most flood tolerant oak and its sound acorns usually float. It is also an attractive shade tree.

Characteristics

Overcup oak grows to a height of 100 feet and a diameter of 5 feet. The bark is light ashy-gray tinged with red under the scales. It occurs in thin, loose scales or broad plates. The 5- to 9-inch-long leaves have five to nine rounded lobes, and they are narrow and wedge-shaped at the base. They are dark green above and pale green beneath. The acorns are about 1 inch in diameter and are rounded or somewhat flattened. The cup is nearly spherical and covers most of the acorn. It is thick at the base and thinner at the margin.

Adaptability

Overcup oak occurs in bottomlands from the coastal plain of Delaware and Maryland to eastern Texas. It is commonly on poorly drained alluvial clay and clay loam soils, but can frequent better sites. In the Mississippi River Alluvial Valley, it occurs on the first bottoms and terraces in and along sloughs and backwater areas. It is adapted to the Southern Mississippi Valley Alluvium and Southern Mississippi Valley Silty Uplands. It is the most flood-tolerant oak and can withstand continuous flooding from January to June.

Establishment

Overcup oak is established by seedlings. The 1- or 2-year-old seedlings are generally of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates:	December 1 to March 31
Plant spacing:	12- by 12-foot (302 trees per acre)
Planting depth:	At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning; those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.8 Swamp chestnut oak (*Quercus michauxii* Nutt.) (QUMI)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

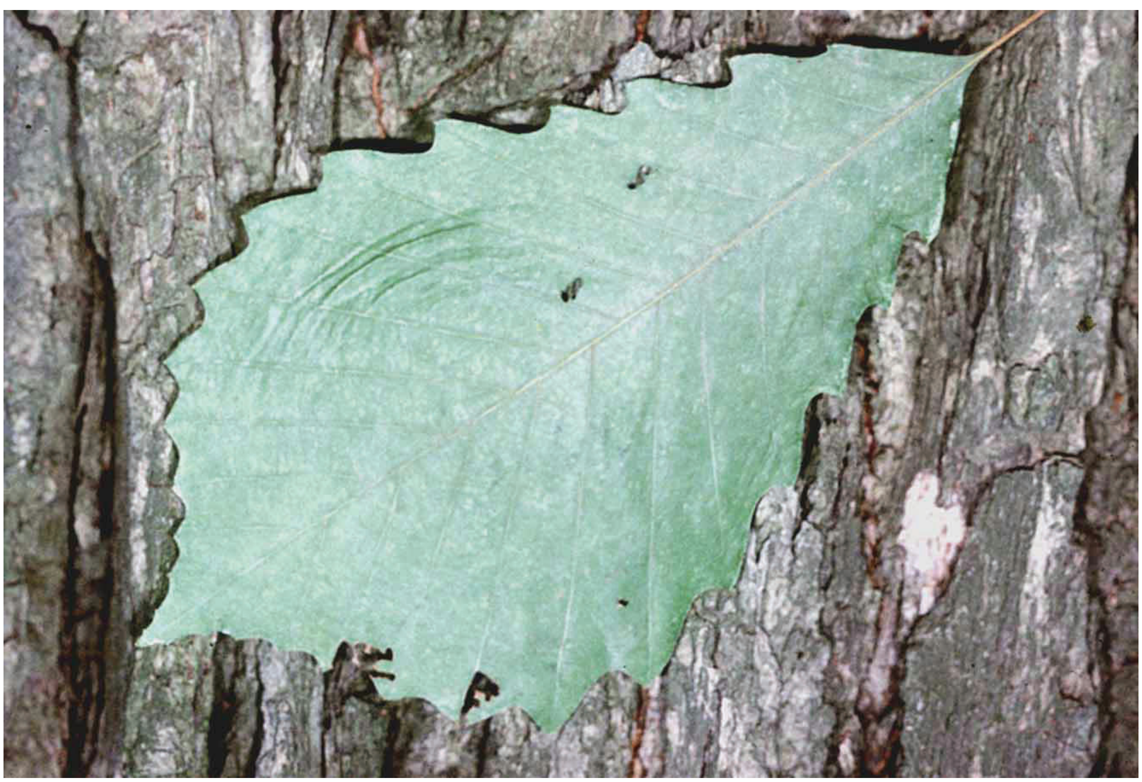
Swamp chestnut oak (also known as basket oak and cow oak) is a member of the white oak group of oaks. It is well adapted to occasionally flooded bottomland areas. It is used for manufacturing lumber, cooperage, veneer, posts, handles, baskets, and fuel. Swamp chestnut oak is highly suited as wildlife food source.

Characteristics

Swamp chestnut oak grows to a height of 100 feet and a diameter of 4 feet. The bark is ashy-gray tinged with red separating into close scales. The 4- to 8-inch-long leaves have shallow adjacent rounded lobes. They are dark green above and soft downy beneath. The acorns are about 1 inch in diameter and 1.5 inches long. The cup is shallow.

Adaptability

Swamp chestnut oak is commonly on moderately well drained, silty clays and loams of first bottoms, ridges, terraces, and colluvial sites along large and small streams. In the Mississippi River Alluvial Valley, swamp chestnut oak is adapted to the Southern Mississippi Valley Alluvium and the Southern Mississippi Valley Silty Uplands. It is tolerant of periodic flooding of up to 2 weeks duration between January and March.



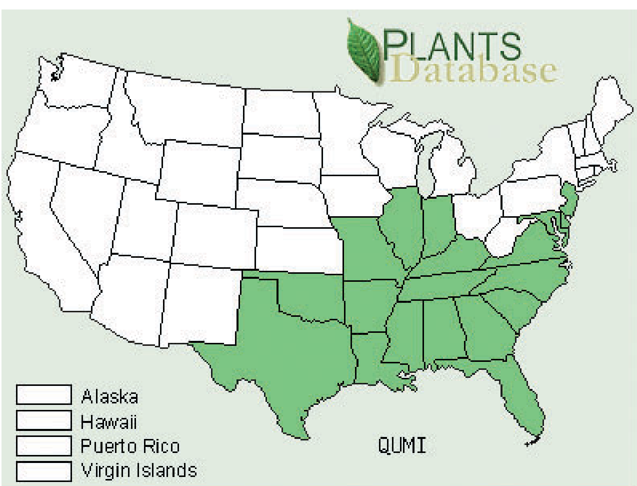
Establishment

Swamp chestnut oak is established by seedlings. The 1- to 2-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 1- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper



Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.9 Water oak (*Quercus nigra* L.) (QUNI)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

Adaptability

Water oak is commonly on moderately well drained loams of first bottoms, terraces, and colluvial sites along large and small streams. It does occur on some upland sites; however, its growth and quality are poor. Water oak is adapted to the Southern Mississippi Valley Alluvium and the Southern Mississippi Valley Silty Uplands. It is tolerant of periodic flooding of up to 3 months duration between January and March.

General use

Water oak is in the red oak group of oaks. It is well adapted to occasionally flooded bottomland areas. It is used for lumber and railroad ties. Water oak produces crops of small acorns and is a fairly consistent mast producer for deer, waterfowl, and turkey. It is also an attractive shade tree.

Characteristics

Water oak is a tree that grows to a height of 100 feet and a diameter of 2 to 3 feet. The bark is moderately thick and tight. It is somewhat ridged; ridges are reddish brown and closely appressed. The leaves are variable in shape, and they are always bristle-tipped (characteristic of all oaks in the red oak group). They are mostly oblong, broader at the outer end, and narrower at the base. They are slightly or distinctly three-lobed at the outer end, thin, dull dark green, paler beneath than above, and mostly smooth. They are 2 to 3.5 inches long and 1 to 1.5 inches wide. The leaves remain green for some time and may persist well into the winter. The acorns are small, round or flattened, and 0.3 to 0.5 inch in diameter. They take 2 years to mature. The cup is shallow and only covers the base of the acorn.

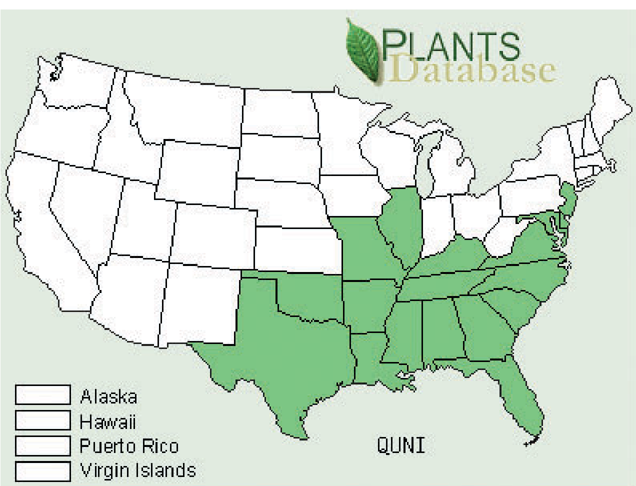
Establishment

Water oak is established by seedlings. The 1- or 2-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper



Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.10 Cherrybark oak (*Quercus pagoda* Raf.) (QUPA5)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph, Norman Melvin; State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

Cherrybark oak is in the red oak group of oaks. It is well adapted to occasionally flooded bottomland area and upland areas with moist soils and northern exposures. It is used for flooring, furniture, construction, sashes and doors, panels, veneer, cooperage, crossties, caskets, and fuel. Cherrybark oak is a fairly consistent mast producer for deer and turkey. It is also an attractive shade tree. Cherrybark oak is the best species for combined timber and wildlife use on occasionally flooded sites.

Characteristics

Cherrybark oak is a tree that grows to a height of 120 feet and a diameter of 4 to 6 feet. The bark is light gray marked with darker bands and somewhat resembling that of black cherry. On the trunk, the bark is dark and rough. The leaves are divided into 5 to 11 sharp-pointed, triangular lobes separated by deep rounded sinuses. They are dark green above and pale and hairy beneath. The leaves are typically 5 to 9 inches long and 4 to 5 inches wide. The rounded acorns are 0.5 inch long. They are borne singly or in pairs and take 2 years to mature. The cup is thin, tight-scaled, and may cover up to a third of the acorn.

Adaptability

Cherrybark oak is commonly on well-drained loams of first bottoms, terraces, and colluvial sites along large and small streams. It also occurs in nonstream associated areas on moist, well-drained sites along the Coastal Plain, such as moist flats and edges of depression wetlands. Cherrybark oak is adapted to the Southern Mississippi Valley Alluvium and the Southern Mississippi Valley Silty Uplands. It is tolerant of periodic flooding of up to 2 weeks duration between January and March.

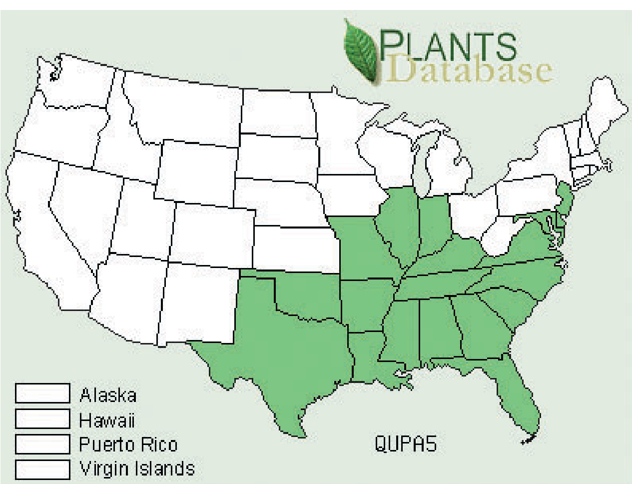
Establishment

Cherrybark oak is established by seedlings. The 1- to 2-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper



Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

Notes

Some older references refer to cherrybark oak as a variety of southern red oak as *Q. falcata* var. *pagodifolia.* It is, however, now considered a distinct species from the southern red oak that occurs in drier habitats.

IV.E.11 Pin oak (*Quercus palustris* Muenchh.) (QUPA2)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

Pin oak is a red oak well adapted to occasionally flooded bottomland areas. It is used for lumber and railroad ties. Pin oak is a fairly consistent mast producer for deer, turkey, and waterfowl. It is also a frequently used shade tree on wet soils.

Characteristics

Pin oak is a tree that grows to a height of 80 feet and a diameter of 3 feet. The bark on young trees is light brown, smooth, and shiny. On older trees, it is tight and smooth, gray brown to black, and covered with small, closely attached scales.

The leaves are deeply lobed, bristle-tipped, lustrous dark-green above and pale beneath. They are typically 4 to 5 inches long and 2 to 3 inches wide. The acorns are small, round, and 0.5 inch in diameter. They are borne singly or in pairs. The acorns mature in 2 years. The cup is shallow and only covers the base of the acorn.

Adaptability

Pin oak occurs on wet sites primarily in the mid-Atlantic and central regions. It occurs on heavy soils with poor drainage, but also occurs commonly on moderately well drained clays and clay loams. Pin oak is adapted to the Arkansas Valley and Ridges, the Southern Mississippi Valley Alluvium, and the Southern Mississippi Valley Silty Uplands (north of Memphis). It is tolerant of periodic flooding of up to 3 months duration between January and May.

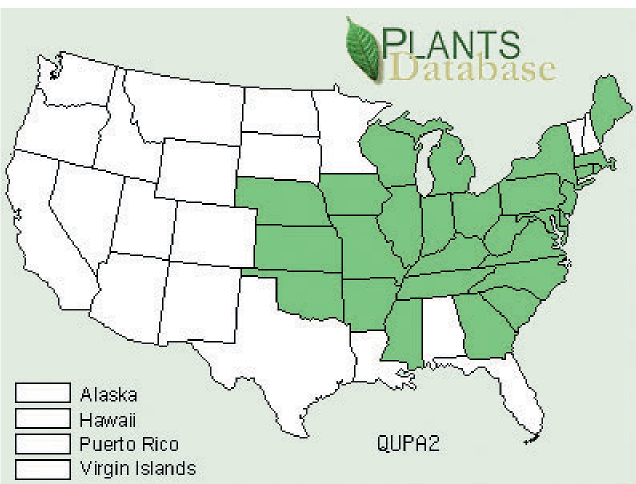
Establishment

Pin oak is established by seedlings. The 1-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper



Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.12 Willow oak (*Quercus phellos* L.) (QUPH)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

Willow oak is in the red oak group of oaks. It is well adapted to occasionally flooded bottomland areas. It is used for agricultural implements, construction, and cooperage. Willow oak is an especially valuable wildlife food source because it is a good mast producer and is the only oak whose foliage is a highly preferred food for deer. It is also an attractive shade tree.

Characteristics

Willow oak is a tree that grows to height of 100 feet and a diameter of 3 feet. In the open, it retains its lower branches, which spread widely. The bark on young trees is thin and smooth. On old trunks, it is broken into shallow, narrow fissures and irregular plates.

The leaves do not have lobes. They are 2 to 3 inches long and 0.25 to 0.75 inch wide with smooth or slightly wavy margins and bristle-points. They are smooth, green, and shiny above, and duller beneath. The acorns are 0.25 to 0.75 inch in diameter. They are borne singly or in pairs. They mature in 2 years. The cup is shallow and red and only covers the base of the acorn.

Adaptability

Willow oak is commonly on moderately well drained silty clays and loams. In the Mississippi River Alluvial Valley, it is adapted to the Southern Mississippi Valley Alluvium, Southern Mississippi Valley Silty Uplands, and the Arkansas Valley and Ridges. It is tolerant of periodic flooding of up to 3 months duration from January to May.

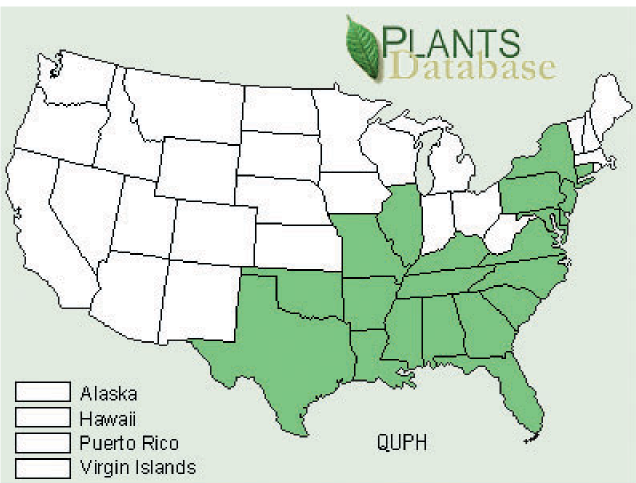
Establishment

Willow oak is established by seedlings. The 1-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper



Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.13 Shumard oak (*Quercus shumardii* Buckl.) (QUSH)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

Shumard oak is in the red oak group of oaks and is well adapted to occasionally flooded bottomland areas and upland areas with moist soils and northern exposures. It is used for flooring, furniture, construction, sashes and doors, panels, veneer, cooperage, railroad ties, caskets, and fuel. Shumard oak is a fairly consistent mast producer for deer and turkey. It is also an attractive shade tree.

Characteristics

Shumard oak is a tree that grows to a height of 100 feet and a diameter of 3 to 4 feet. The bark on young trees is smooth and light gray to reddish brown. On older trees, it is dark brown tinged with red and is broken by shallow fissures into regular, flat, and usually smooth plates or strips. The leaves are divided into seven to nine rounded, bristle-tipped lobes. They are dull green above and pale beneath. They are typically 5 to 9 inches long, 4 to 6 inches wide, and broader toward the tip. The blunt-tipped, flat-based acorns are 0.75 to 1.5 inches long. They take 2 years to mature. The cup is flat, shallow, and dark brown.

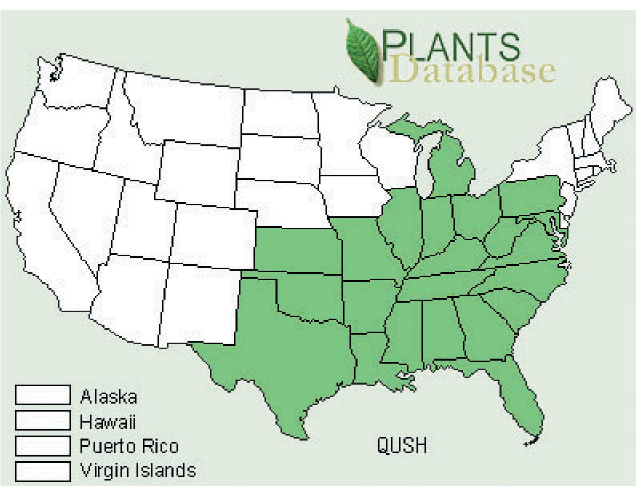
Adaptability

Shumard oak is commonly on well-drained loams of terraces, colluvial sites, and bluffs adjacent to large and small streams. It is adapted to the Southern Mississippi Valley Alluvium and the Southern Mississippi Valley Silty Uplands. This oak is tolerant of periodic flooding of up to 2 weeks duration between January and March.

Establishment

Shumard oak is established by seedlings. The 1-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates:	December 1 to March 31
Plant spacing:	12- by 12-foot (302 trees per acre)
Planting depth:	At depth in nursery to an inch deeper



Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.14 Texas red oak (*Q. texana* Buckl.) (QUTE)

synonym: Nuttall oak
(*Quercus nuttallii* Palmer)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; photograph and State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana)

General use

Texas red oak (also commonly referred to as nuttall oak) is in the red oak group of oaks. It is well adapted to frequently flooded bottomland areas. This oak is used for flooring, furniture, construction, sashes and doors, panels, veneer, cooperage, railroad ties, caskets, and fuel. Texas red oak is an especially valuable wildlife food source because its acorns fall gradually throughout the winter. Deer, turkey, and waterfowl use Texas red oak acorns. It is also an attractive shade tree.

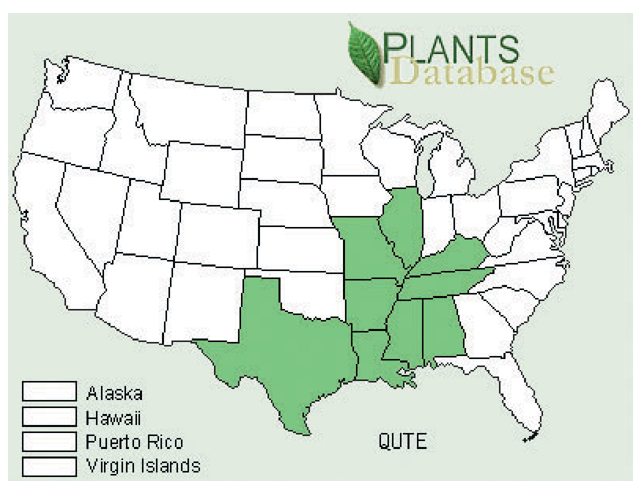
Characteristics

Texas red oak is a slender tree with a conical crown that grows to a height of 75 feet and a diameter of 4 feet. Its upper branches are ascending, and lower branches are horizontal or drooping. The bark on young trees is olive-green tinged with red or reddish-brown. On larger branches, it is gray to dark brownish-gray; on old trunks, it is dark, slightly fissured, and relatively smooth.

The leaves are similar to pin and shumard oak and have five to seven lobes at nearly right angles to the mid-vein. The middle or upper pair of lobes is the longest. Each lobe is slightly tapered to the end and terminates in bristle-tipped teeth. The acorns are about 1 inch long, and 0.5 to 0.75 inch in diameter. The cup is deep and covers a third to a half of the acorn. It tapers abruptly downward into a short, scaly stalk; the scales are slightly hairy. The acorn is dark brown with lighter colored vertical stripes.

Adaptability

Texas red oak is commonly on poorly, drained alluvial clay soils. It is less commonly on silty clay flats and sloughs on the terraces of major streams. This oak is adapted to the Southern Mississippi Valley Alluvium and the Southern Mississippi Valley Silty Uplands. It is one of the most flood-tolerant oaks and can withstand continuous flooding from January to May.



Establishment

Texas Red Oak is established by seedlings. The 1-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates: December 1 to March 31

Plant spacing: 12- by 12-foot (302 trees per acre)

Planting depth: At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

IV.E.15 Bald cypress (*Taxodium distichum* (L.) L.C. Rich.) (TADI2)

(Nancy Young, USDA-NRCS Arkansas, and Bob Glennon, USDA-NRCS (currently USFWS), authors; Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, edited December 2001; State distribution map, NRCS Plants Data Center, Baton Rouge, Louisiana; photograph, Jody Pagan, NRCS, Little Rock, Arkansas)

General use

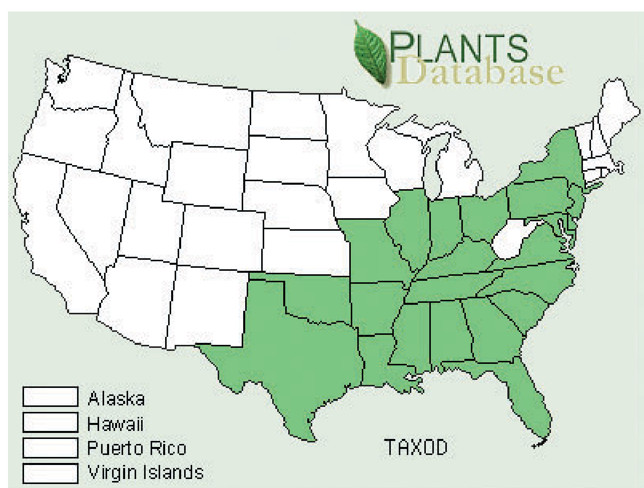
Bald cypress is well adapted to frequently flooded bottomland areas. It is used for general construction, boat building, fence posts, poles, pilings, shingles, railroad ties, silos, caskets, and cooperage. Bald cypress is suited as wildlife food source for small birds and a nesting and perch site for riparian bird populations. It is also an attractive shade tree.

Characteristics

Bald cypress grows to a height of 120 feet and a diameter of 10 feet at the buttressed roots. It has a conical shape in youth; in old age, it has a broad, open top of a few heavy and numerous small branches. The bark is cinnamon red to silvery gray and is finely divided by numerous longitudinal fissures. It peels off in long strips on older trees. The leaves are flat and needle-like. They are 0.5 to 0.75 inch long in two rows. The leaves and the small twigs to which they are attached fall off in the autumn. The fruit is a rounded ball about 1 inch in diameter with thick, irregular scales.

Distribution and adaptability

Bald cypress is adapted (natively) to very wet soils. It occurs on poorly drained alluvial clays and on clay loams. This tree is tolerant of continuous flooding from January to June. Although the species has been recorded as occurring in most States from New York to Texas, it generally ranges along the Southeast Atlantic and Gulf Coastal Plain and up the Mississippi River Alluvial Valley into southern Illinois and Indiana. In the Lower Mississippi Alluvial Valley, it occurs on Southern Mississippi Valley Alluvium, Southern Mississippi Valley Silty Uplands, and the Arkansas Valley and Ridges.



Establishment

Bald cypress is established by seedlings. The 1-year-old seedlings are usually of adequate size for planting. Sites must be prepared as necessary to facilitate good survival. Land that has been used recently to produce an annual crop may not need any preparation. Land that has succeeded to annual weeds may need to be mowed to facilitate machine planting. Land that has stands of perennial grasses and legumes must be sprayed or tilled at least in strips where the trees will be planted. Land that has succeeded to brush must be treated by mechanical or chemical means to control the plants. Plantings in existing woodland must have unwanted overstory trees removed before planting.

Planting dates:	December 1 to March 31
Plant spacing:	12- by 12-foot (302 trees per acre)
Planting depth:	At depth in nursery to an inch deeper

Management

Protect the young trees from fire and harmful grazing. Grazing should be excluded from the stand where canopy is less than 12 feet in height. Fire protection is necessary at all times.

The stand should be evaluated at the age of 20 to 25 years for thinning needs. The trees should be reaching a point where the canopy is closing and causing slow growth rates. A thinning at this point stimulates growth and seed production. A spacing guide using the average tree diameter in inches times 1.75 will maintain the proper spacing in feet.

The least desirable trees should be removed first during a thinning: those with poor form, injuries, or low vigor. After that, remove additional trees until an adequate spacing is achieved. Extreme care should be taken during the harvest operation to avoid damage to the remaining trees. Injuries from skidding and other cutting activities increase the incidence of rot and degradation of the stand.

The optimum interval between thinnings depends upon the capability of the soil and the restoration objectives. A rule-of-thumb is to examine the stand every 7 to 10 years for crown closure and slowed growth from a wildlife perspective. With wetland restorations and enhancements intended to provide or maximize wildlife habitat, care should be taken not to remove the number or size of trees during the thinning process that would have an adverse impace on targeted species. Generally, tree removal should be based upon wildlife needs as opposed to economic consideration.

Notes

A similar, but distinct species, pondcypress (*Taxodium ascendens*), also occurs in the South and Southeast. Some references consider both of these species one-and-the-same, but pondcypress is considered a species separate from bald cypress both in appearance (taxonomic) and habitat. Pondcypress generally occurs in bay heads, ponded Carolina bays, and upland swamp forests as opposed to alluvial swamps where bald cypress commonly occurs. Consider the restoration site, and select the appropriate species.

IV.F Selection of herbaceous vegetation for moist-soil management areas in the Midwest, South, and Southeast

(Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001)

Purpose

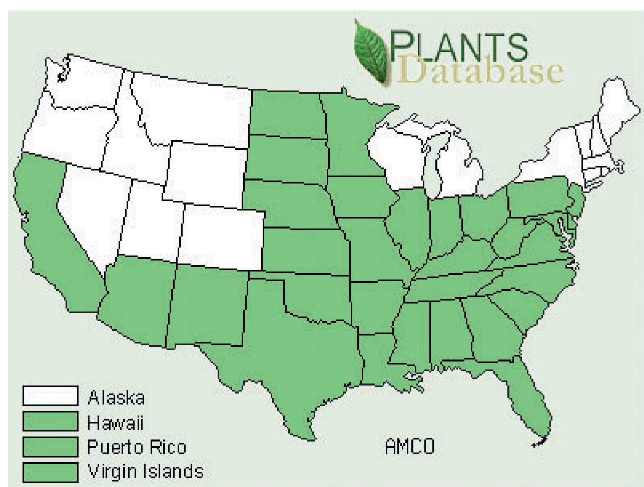
The purpose of this section is to provide information on selected herbaceous plant species that occur and can be encouraged to enhance the wildlife benefit in moist-soil management areas in the Midwest, South, and Southeast. These species are being used in wetland restoration and enhancement projects and are recommended for use (within limitations noted). Each plant sheet in this section contains information on the plants general economic and wildlife use, identification characteristics, habitat and geographic distribution, establishment, and management techniques. Generally, the group is considered with multiple beneficial species.

Contents

The following species are included in this section:

IV.F.1 Toothcup (*Ammannia coccinea* Rottb.)

(Text by Mark Tidwell and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photograph and distribution map by NRCS Plants Data Center, Baton Rouge, Louisiana)



Toothcup (valley redstem or purple ammannia) is a native annual herb associated with moist to wet sites in moist-soil impoundments. Toothcup grows up to 2 feet high, has square stems, and slender, linear, opposite leaves. They are early successional species that germinate late in the growing season. Best seed production occurs following late spring or summer drawdowns. Normally occurs with sprangletop.

Plant value

Toothcup is a valuable source of seeds for gadwall and pintails. Although the individual seeds are small (600,000 seeds/lb), seed production may be as high as 500 pounds per acre. The plants are also considered as having a high palatable browse potential.

Frequency of occurrence

Problem:	Never a problem (in moist-soil management areas.)
Severe problem:	Never a severe problem (in moist-soil management areas.)

Enhancement

Maintaining vegetation in early successional stages and lengthening the period soils are in a moist condition increase germination of toothcup. High seed production always is associated with a summer drawdown or on drier sites in wet years or wet sites in dry years. Therefore, periodic mechanical disturbances (i.e., shallow disking) and irrigation treatments often can be used to enhance toothcup occurrence and seed production.

Notes

Although toothcup is not considered a problem from a moist-soil management perspective, it is listed as an invasive species in one source: Southern Weed Science Society. 1998. Weeds of the United States and Canada. CD-ROM, Southern Weed Science Society, Champaign, Illinois.

IV.F.2 Bidens, beggarticks, sticktights, boot jacks, tickseed sunflower, Spanish needle (*Bidens* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photographs by William S. Justice and NRCS Plants Data Center, Baton Rouge, Louisiana)

Bidens is a common group of plants in moist-soil impoundments. There are more than 10 species of Bidens that occur in wetlands within the United States, most of which are native and annual. Characteristic of early stages, bidens are dicots and members of the Compositae family. Germination is usually associated with dry to moist soil conditions. Best germination and seed production occurs following late spring or summer drawdowns. Some species germinate early, but the bright yellow flowers do not appear in most species until late summer or early fall. They are prolific seed producers.

Plant value

Bidens plants are excellent seed producers, and seeds are readily consumed by mallards. The seeds are particularly high in protein. Listed here are a few common species.

- Bearded beggarticks (*Bidens aristosa* (Michx.) Britt.) is a robust plant with peak flowering in September. Seeds tend to drop readily from the plants. Weeds of Kentucky and adjacent states: A field guide, 1991, by P.D. Haragan lists this species as an invasive. (*Bidens polylepis* is currently considered a synonym of *B aristosa.*)
- Devil's beggarticks (*B. frondosa* L.) is a robust plant with peak flowering in September. It occurs in wetlands in all the lower 48 states except Montana. Although this species is native to the United States, it is listed by several references as an invasive species. Consideration should be given to this invasive potential when encouraging this species in a moist-soil management area.

Bearded beggarticks

Devil's beggarticks

- Nodding beggarticks (*B. cernua* L.) is of smaller stature and more tolerant of wetter conditions than others listed here. It has been listed by one reference source as an invasive species in the Northern Plains.
- Threelobed beggarticks (*B. tripartita* L.) (*B. comosa* is a synonym) is a low growing form with compact seed heads. This species is more prone to late season germination. Ducks clip entire heads while foraging if seeds are still attached.

Frequency of occurrence

These species are never considered a problem in moist-soil management areas because of their seed production and are to be encouraged. Since there are many species, it would be helpful to know of those that occur in your area. Care should be given in the encouragement of the species since many are considered invasive in some parts of the United States. Consult with State agencies concerning the invasive listing of species in your area. Also refer to the USDA NRCS- Plant Data Center on noxious and invasive species (**http://plants.usda.gov**).

Nodding beggarticks

Threelobed beggarticks

IV.F.3 Sedges (*Carex* spp.)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photographs by NRCS Plants Data Center, Baton Rouge, Louisiana)

Fringed sedge

The *Carex* genus of **sedges** exhibit a cosmopolitan distribution with an estimated 2,000 species worldwide. Similarly, they represent the largest genus of flowering plants in North America with more than 500 species. They are characteristic of later successional stages. Some sedges are robust perennials with rhizomes, whereas others are annuals with fibrous root systems. With so many species, germination requirements are variable. Some species are capable of growth on dry ground, but most species occur in areas with moist to wet soils.

Plant value

Sedges are valuable as rail habitat, providing both robust vertical cover that withstands flooding and seeds that are heavily consumed. Waterfowl consume sedge seeds in moderate amounts. Production of fox sedge (*C. vulpinoidea*) approaches 180 pounds per acre in some years.

Frequency of occurrence

Problem:	>65 percent cover
Severe problem:	Never a severe problem

Control strategies

The strategies for control of sedges are agriculture, burn, deep disk, late disk then flood, mow, mow then semipermanent, semipermanent, plow, shallow disk, herbicide, ignore. Because sedges provide important rail habitat, control should be initiated only if the density of sedges hinders seed production of more desirable plants or a mix of undesirable species (e.g., broomsedge) are found in association with sedges. To maximize the value of this species, control actions should be timed to permit use of these habitats by rails prior to rehabilitation.

Wheat sedge

Gray's sedge

IV.F.4 Chufa (*Cyperus esculentus* L.)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photograph and distribution map by NRCS Plants Data Center, Baton Rouge, Louisiana)

Chufa, chufa flatsedge, or yellow nutsedge is a perennial that rarely produces seed, but consistently produces underground nutlets of great value as a waterfowl food. The plant is widespread across the United States. Generally, the plant develops early in the growing season and senesces in July in Missouri. Little evidence of the aboveground structure remains by the time of fall flooding.

Plant value

The underground nutlet is the most important food produced by this plant. The small seeds are never produced in abundance.

Invertebrates

Chufa has limited value as litter for invertebrates in most situations because the aboveground biomass is largely decomposed before fall flooding. In cases where chufa is a late-season plant, the structure of the plant provides moderately good cover for invertebrates.

Frequency of occurrence (as a moist-soil plant)

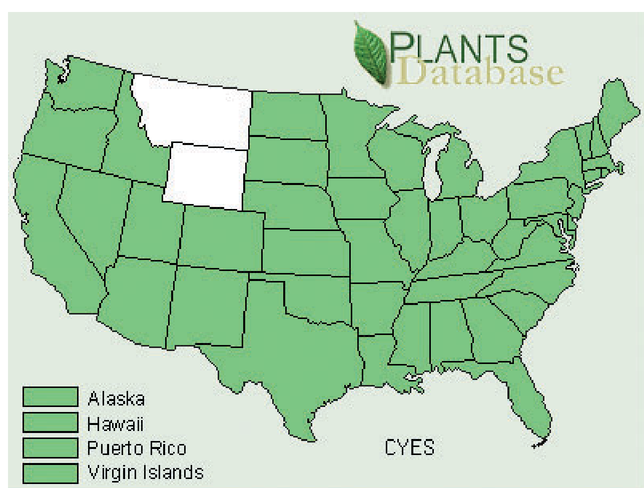
Problem: Never a problem
Severe problem: Never a severe problem

Enhancement

Shallow disking early in the growing season often results in greater stem densities of this plant. Quite often the parent plant is not killed and begins to grow.

Noxious weed information

Chufa flatsedge is valuable as a wildlife plant in moist-soil management areas. Caution is advised when encouraging this plant in that it has noxious and invasive potential. Chufa is on the list of noxious species in the following states: California, Colorado, Hawaii, Minnesota, Oregon, and Washington.



IV.F.5 Flatsedge, umbrella sedge, nutsedge (*Cyperus* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photographs and distribution maps by NRCS Plants Data Center, Baton Rouge, Louisiana)

Flatsedges can be subdivided into two main groups: perennials and annuals. Perennial flat sedges are herbs with creeping rhizomes that may produce tubers, an important waterfowl food. For information on perennial umbrella sedges, see chufa (*C. esculentus* L.) in this section. Annual umbrella sedges, which include redroot flatsedge (*C. erythrorhizos* Muhl.) and ricefield flatsedge (*C. iria* L.), reproduce by seeds and do not have rhizomes and tubers. Early to late spring drawdowns generally tend to result in the best seed production of ricefield flatsedge, whereas late spring to summer drawdowns result in the best seed production of redroot flatsedge. Slow drawdowns lasting longer than 2 weeks tend to increase the germination density of both species because they are adapted to wetter soil conditions. Redroot flatsedge germinate in fresh to slightly brackish water. The salt tolerance of ricefield flatsedge has not been documented. In areas with saline water conditions, the potential for increasing salt loads in the impoundments should be an important consideration in determining the drawdown date and rate used to promote annual flatsedges.

Plant value

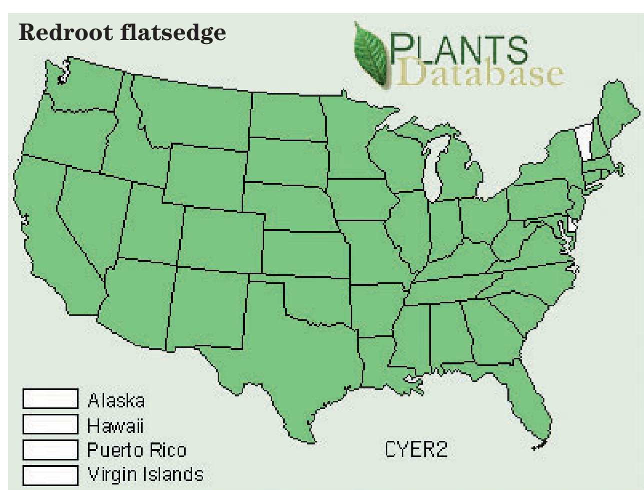
Rails and a variety of waterfowl readily consume seeds of annual flatsedge. Although seeds are small, high densities often accumulate in small areas as they are windrowed against standing vegetation, making them readily available to species ranging from teals to mallards. Seed production of redroot flatsedge and ricefield flatsedge may reach 2,000 pounds per acre and 160 pounds per acre, respectively, during early successional stages.

Frequency of occurrence

Problem: Never considered a problem (in moist-soil management areas)

Severe problem: Never considered a problem (in moist-soil management areas)

Redroot flatsedge



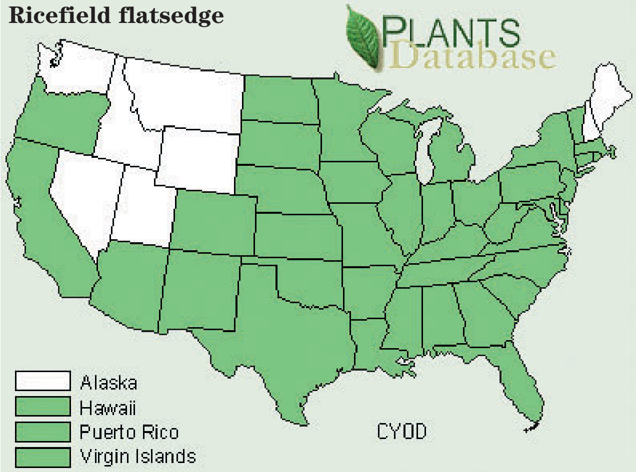
Enhancement techniques

Disking that creates a finer seedbed enables a higher density of the small annual flatsedge seeds to germinate. In some cases cross disking or use of a cultipacker after disking enhances germination to an even greater extent. In dry years irrigation often helps promote higher seed production.

Notes

Redroot flatsedge and ricefield flatsedge have been listed in at least one authoritative reference (Southern Weed Science Society. 1998. Weeds of the United States and Canada. CD-ROM, Southern Weed Science Society, Champaign, Illinois) as a potentially invasive species. Neither of these species is formally listed on any State or Federal list as noxious or invasive. In addition, ricefield flatsedge is not a native species to the United States. It is an introduction from Eurasia. When encouraging these species in moist-soil management areas, it may be important to consider both the potential invasive nature and the non-native status.

Ricefield flatsedge



Ricefield flatsedge

IV.F.6 Crabgrass (*Digitaria* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photograph and distribution map by NRCS Plants Data Center, Baton Rouge, Louisiana)

Crabgrasses are members of the Paniceae tribe of the Gramineae family. They are low growing annuals and are most common on drier moist-soil sites. Best germination and seed production occurs following late spring or summer drawdowns. This is when ambient air temperatures are moderate to high and the soil dries at a sufficient rate to permit crabgrass to become established before the germination and early growth of more robust plants adapted to wetter sites. Normally, other seed-producing species occur in association with crabgrass and contribute to the overall seed production in an impoundment.

Plant value

Seed of crabgrass are consumed by rails and many waterfowl species, particularly teals and pintails. Seed production may reach 200 pounds per acre.

Frequency of occurrence

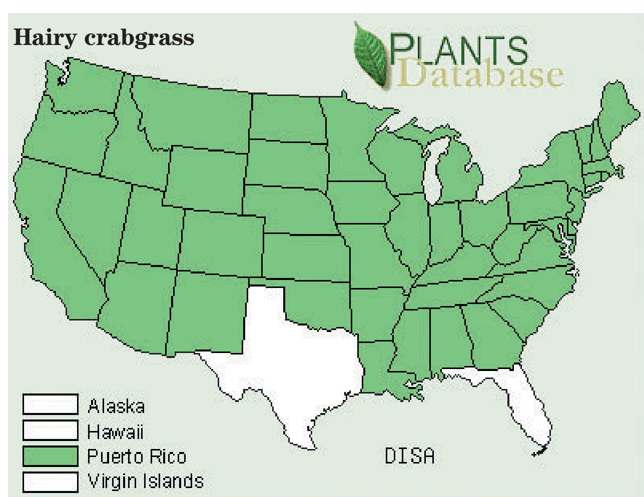
Problem: Never a problem in moist-soil areas

Severe problem: Never a severe problem in moist-soil areas

Enhancement

Crabgrass is never considered a problem in moist-soil management areas and often represents a second source of seed produced in an impoundment. Other species stimulated by late spring or summer drawdowns, such as millet, often occur in conjunction with crabgrass if soil moisture levels are favorable. Mowing taller, undesirable vegetation prior to crabgrass seed formation may enhance crabgrass seed production. This increases sunlight penetration and reduces competition for moisture and nutrients. Care must be exercised to ensure that the apical meristem of favorable plants (e.g., millet) that have already initiated seed set are not destroyed by mowing.

Hairy crabgrass



Notes

Some crabgrass species are listed in authoritative references as being invasive. For example, the common hairy crabgrass (*Digitaria sanguinalis* (L.) Scop.) is listed as invasive in the Northeast, Nebraska and the Great Plains, the West, and Kentucky. It is also a problem yard weed grass. It, however, is not listed on any State or Federal list of noxious or invasive species. Consider these factors when encouraging or enhancing the proliferation of these species.

IV.F.7 Millet/barnyard-grass/watergrass (*Echinochloa* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photographs by NRCS Plants Data Center, Baton Rouge, Louisiana)

Millets are a group of annual grasses that have high food value for waterfowl across the continent. Seeds require moist to saturated soils for germination. In general, drawdowns conducted during late spring result in the best seed production. Some species respond better following early spring drawdowns, and others respond better following summer drawdown. Millets grow on a variety of soils, are readily digestible, and the seeds decompose very slowly when subjected to prolonged flooding.

Characteristics of different species

Billion-dollar grass (*Echinochloa frumentacea* Link.)
This species, introduced from Eurasia, is identifiable from the other millets in that its flowering branches are not widely spreading. Best response for seed production occurs with an early drawdown. Sometimes this species also responds to late drawdowns, but stems generally are widely scattered.

Billion-dollar grass

Japanese millet or barnyard grass (*Echinochloa crusgalli* (L.) Beauv.)
Best seed production occurs with a midseason drawdown. Under some conditions, however, seed production following a late season drawdown is as good as production following a midseason drawdown. Response is poor with an early drawdown. Seeds germinate in brackish water, but seedlings do not survive salinity exceeding 5-ppt total dissolved solids. This species is also introduced from Eurasia, but differs in appearance from *E. frumentacea* in that its (*E. crusgalli*) flowering branches are more spreading. Although no State has placed this species on its invasive species list, six authoritative references on weeds indicate that it is potentially invasive in most of the United States. For more information consult the NRCS PLANTS Database (http://plants.usda.gov).

Barnyard grass

Rough barnyard grass (*Echinochloa muricata* (Beauv.) Fern.)
Best response for seed production occurs with a late season drawdown. Midseason drawdowns also produce good results. This introduced species is similar in appearance to the other millets listed, but it can be identified by the presence of small bumps (i.e., muri) on the chaff (glumes and lemmas).

Coast cockspur grass (*Echinochloa walteri* (Pursh) Heller)
This native species seems to do best where soils are a silt loam. Response is good following mid and late season drawdowns. This is the most tolerant of wet conditions of all the millets listed. It can be distinguished from the other species in that it has spikelets about three times as long as broad and it has conspicuously long awns.


Coast cockspur grass

Plant value

- **Excellent seed producer**—During the first growing season after soil disturbance, seed production may reach 1,500 to 3,000 pounds per acre. Seed production gradually decreases in subsequent years as follows: year 2, 1,200 to 2,000 pounds per acre; year 3, 800 to 1,200 pounds per acre.
- **Invertebrate substrate**—Millets provide a moderately valuable substrate for invertebrates.

Enhancement techniques

Slow drawdowns, particularly during midseason, generally produce excellent results. Soil disturbance after three or four seasons increases production. Best production generally occurs when the soil is disturbed late in the previous growing season. A good technique to increase millet in the next growing season consists of disking in late summer or early fall followed by shallow flooding to provide shorebird habitat.

Control

Control within moist-soil management areas is never considered necessary for millets.

IV.F.8 Spikerush (*Eleocharis* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photographs by NRCS Plants Data Center, Baton Rouge, Louisiana)

Spikerushes are a group of sedges that are characterized by having a cone-like inflorescence on the end of its stems. Numerous species of spikerushes occur in wetlands and moist-soil management areas; however, not all of them are of equal value or beneficial to wildlife. Several of the common beneficial and not so beneficial species are listed.

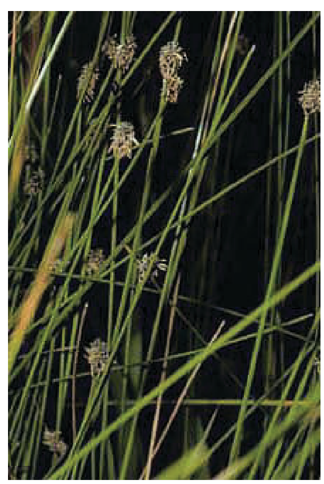
Blunt spikerush (*Eleocharis obtusa* (Willd.) J.A. Schultes)
Blunt spikerush is a small, annual rush that provides a high quality green browse. The plant is widespread throughout the Midwest and often occurs as a carpet under more robust moist soil vegetation. Although germination is most prolific following late winter drawdowns, blunt spikerush is capable of germinating throughout the growing season. This is one of the first colonizers of newly restored wetlands in the mid-Atlantic region.

Blunt spikerush

Least spikerush (*E. parvula* (Roemer and J.A. Schultes) Link ex Bluff)
Least spikerush (also known as dwarf spikerush) is a short, mat-forming, annual spikerush common in Atlantic coastal areas. It may form extensive stands on wet saline soils. The filiform stems often are yellow or brownish. Germination is best following early spring drawdowns.

Least spikerush

Common spikerush (*E. palustris*) and squarestemmed spikerush (*E. quadrangulata*)
The round-stemmed common spikerush and the larger squarestemmed spikerush are problematic and tend to become increasingly common on moist-soil sites after several years of management. Their large size and perennial habit makes them of poor value as a browse, and the small seeds are relatively unimportant as a food source.



Common spikerush

Squarestemmed spikerush

Plant value

Greatest value of the small, annual spikerushes is as a green browse. The plants do produce an abundance of very small seeds, but the seeds are rarely detected during food habitat studies. The best technique to enhance browse production is to disk and then follow by irrigation to bring soil to saturation. This technique works particularly well in late summer or early fall to provide browse for geese.

The more robust, perennial species may have limited value as a protective cover in the early stages of establishment, but once established they tend to create a thick clump.

Control

The annual spikerushes are never considered a problem; thus, control is not necessary.

The perennial species are considered a problem with the density between 10 and 25 percent in scattered clumps. A severe problem exists when the cover exceeds 20 percent in dense clumps or if the site has more than 40 percent cover total.

Control strategies

The best long-term strategy is to keep large clumps of perennials from developing. This can be accomplished by varying the hydrologic regime among years or continuously setting back succession using soils disturbance techniques (disk followed by drying, deep disking, burning, plowing).

Notes

Recent changes in nomenclature have combined several species of Eleocharis together. *Eleocharis smallii* and *E. macrostachya* are currently considered synonyms of *E. palustris*. Also the perennial common spikerush (*E. palustris*) is similar in appearance to the annual blunt spikerush (*E. obtusa*) and generally follows it in a succession sequence. Knowledge of the two species identification is important so that it can be determined when the annual species gives way to the less valuable perennial species.

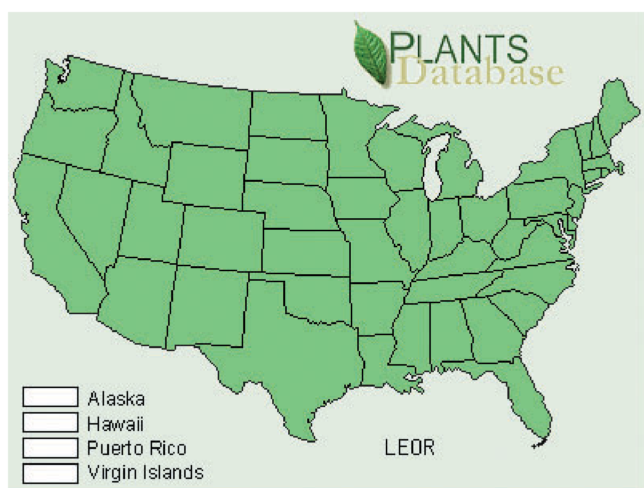
IV.F.9 Rice cutgrass (*Leersia oryzoides* (L.) Sw.)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photograph and distribution map by NRCS Plants Data Center, Baton Rouge, Louisiana)

Rice cutgrass is a perennial grass with creeping rhizomes. A late successional species adapted to moist or wet sites, best germination and seed production occurs following late spring and summer drawdowns. Reproduction primarily is by seeds.

Plant value

Rice cutgrass is a valuable source of seeds for numerous waterfowl species. Rootlets appear in the late winter/early spring diets of mallards. In addition, vegetative parts also serve as an invertebrate substrate. On recently disturbed sites, rice cutgrass is not common and seed production may approach only 45 pounds per acre. However, seed production can exceed 325 pounds per acre on sites that have not been recently disturbed and are dewatered late in the growing season.



Frequency of occurrence

Problem: Never a problem

Enhancement

Implementing a slow drawdown late in the growing season can increase germination of rice cutgrass. The objective is to maintain high soil saturation for as long as possible.

IV.F.10 Bearded sprangletop (*Leptochloa fusca* ssp. *fascicularis* (Lam.) N. Snow) and Mucronate sprangletop (*Leptochloa panicea* ssp. *brachiata* (Steudl.) N. Snow)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photograph by NRCS Plants Data Center, Baton Rouge, Louisiana)

Sprangletops are annual grasses associated with moist to wet sites in moist-soil impoundments. They are early successional species that germinate late in the growing season. Best seed production occurs following summer drawdowns.

Bearded spangletop

Plant value

Sprangletop is a valuable source of seeds and provides cover for waterfowl. In addition, vegetative parts also serve as an invertebrate substrate. Seed production can exceed 1,500 pounds per acre in recently disturbed sites that are dewatered late in the growing season. Seed production is lower (300 lb/ac) in areas that are dewatered early in the season. Regardless of drawdown date, production gradually decreases in subsequent years following disturbance.

Frequency of occurrence

Problem: Never a problem

Enhancement

Maintaining vegetation in early successional stages and lengthening the period soils are in a moist condition increase germination of sprangletop. High seed production always is associated with a summer drawdown or on dryer sites in wet years or wet sites in dry years. Therefore, periodic mechanical disturbance (i.e., shallow disking) and irrigation treatments often can enhance sprangletop occurrence and seed production.

Notes

Recent taxonomic changes to the scientific names of these sprangletops have lead to some confusion as to the correct scientific name. The species formerly known as L. filiformis is now considered only a subspecies of bearded sprangletop: *Leptochloa fusca* ssp. *fascicularis* (Lam.) N. Snow. Similar name changes have occurred with mucronate sprangletop. *Leptochloa fascicularis* is considered as a subspecies of *L. panicea* as ssp. *brachiata* (Steudl.) N. Snow.

IV.F.11 Panicgrasses (*Panicum* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photographs by NRCS Plants Data Center, Baton Rouge, Louisiana)

Panicgrasses are characteristic of early successional stages. Common on drier moist-soil sites, germination and seed production of these low-growing annuals is best following late spring or summer drawdowns. Normally, other seed producing species occur in association with panicgrass and contribute to the overall seed production in an impoundment.

Plant value

Seeds of panicgrasses are consumed by rails and many waterfowl species, particularly teals and pintails. Seed production may be as high as 400 pounds per acre in years immediately following soil disturbance. In subsequent years, seed production gradually decreases.

Frequency of occurrence

Problem: Never a problem

Severe problem: Never a severe problem

Enhancement

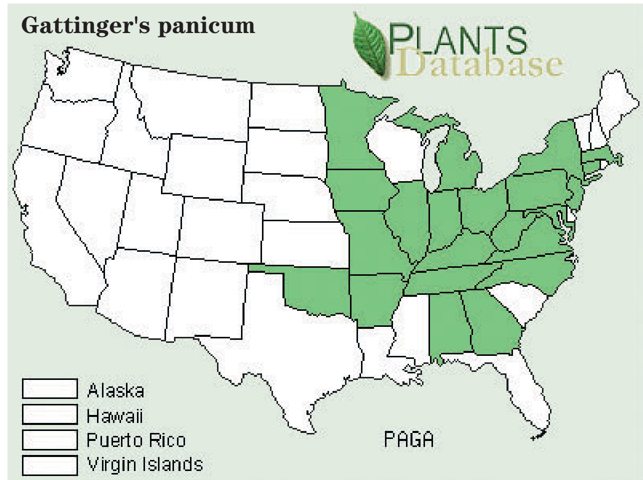
Panicgrass is never considered a problem and often represents a second source of seed produced in an impoundment. Other species stimulated by late spring or summer drawdowns, such as millet and crabgrass, often occur in conjunction with panicgrass if soil moisture levels are favorable at the correct time. Mowing taller, undesirable vegetation prior to panicgrass seed formation may enhance panicgrass seed production. Mowing should occur at a height that removes the apical meristem of undesirable plants, but does not harm desirable plants.

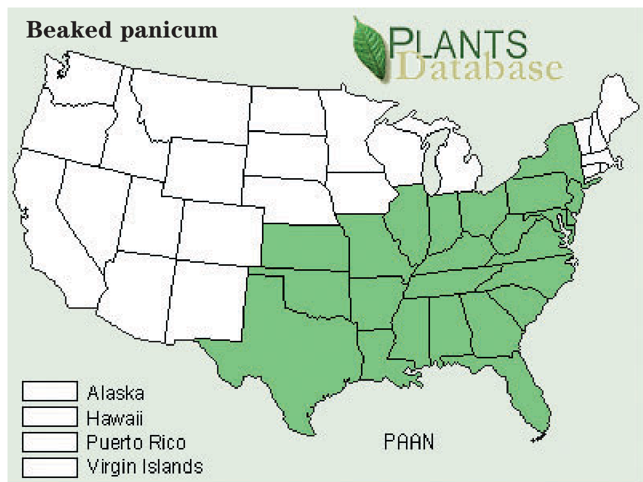
Gattinger's panicum

Beaked panicum

Notes

Recently the panicgrass genus (*Panicum*) has been split into two genera. The genus *Dichanthelium* is perennials that have two distinct growth phases: a winter rosette of leaves and a summer growth phase with leaves dissimilar in appearance from the winter phase. A common example is deertongue (*D. clandestinum*). The genus *Panicum* is both annuals and perennials, but these plants have no overwintering basal rosette of leaves. A common example is switchgrass (*P. virgatum*).





IV.F.12 Smartweeds/knotweed (*Polygonum* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photographs by William S. Justice and NRCS Plants Data Center, Baton Rouge, Louisiana)

Smartweeds and **knotweeds** are widely recognized as a valuable food for waterfowl. Seeds have a hard coat that is not easily broken down in the digestive tract; thus, true metabolizable energy is low for smartweeds. Seeds require moist to saturated soil conditions in freshwater systems for germination. Smartweeds occurring in the United States include native and non-native species, annuals and perennials, as well as species that are adapted to wetlands and those adapted to uplands.

Annual smartweeds

The native annual smartweeds, which include curlytop knotweed (*P. lapathifolium* L), marshpepper knotweed (*Polygonum hydropiper* L.), Pennsylvania smartweed (*P. pensylvanicum* L.), and others are good seed producers, have a wide distribution, and are considered to be of great importance as waterfowl food. These species respond best to drawdowns conducted early in the growing season and normally occur on wetter sites within a unit. Highest seed production typically occurs the year after a soil disturbance or in areas that are drawn down after a period of prolonged flooding. Some annual introduced smartweeds, in particular, chickenweed (or Asiatic tearthumb) (*Polygonum perfoliatum* L.) are not considered beneficial. Chickenweed is invasive and listed as a noxious weed in North Carolina, Ohio, and Pennsylvania.

Plant value

Seed production—Excellent seed production is best the first year after disturbance and may approach 2,000 pounds per acre. Seed production decreases rapidly in each succeeding year and may be only a few hundred pounds per acre by the third year after soil disturbance.

Invertebrate substrate—Excellent substrate for invertebrates, but leaves must remain attached to the stem. Drought and insect infestations reduce leaf abundance and thus invertebrate populations.

Techniques to enhance seed production—Early drawdowns are essential. Soil disturbance (e.g., disking) can be used as a technique to provide shorebird habitat in the same year as the disturbance and increase smartweed seed production the following growing season. Early dewatering of areas that have been deeply flooded for one or more continuous years also result in high seed production.

Control

Controlling native annual smartweeds is never necessary.

Pennsylvania smartweed

Curlytop knotweed

Perennial smartweeds

The two most common perennial smartweeds are water smartweed/knotweed (*P. amphibium* L.) and water pepper (*P. hydropiperoides*). Both species occur in the wetter sites within units. *P. hydropiperoides* usually is associated with sites that have some surface water until July or sometimes even later.

Plant value

Seed production—Seed production is poor, and the seeds are small and hard with poor digestibility. Seed production varies among years, but is never as high as production by annual smartweeds.

Invertebrate substrate—Excellent substrate for invertebrates provided the leaves remain on the stem.

Control

Short-term control strategies involve deep soil disturbance, such as deep disking and plowing. Long-term strategies should involve keeping the impoundment in a dryer condition for at least 2 years.

Frequency of occurrence

Problem: > 30%
Severe problem: > 30% in a solid block or > 60% as scattered clumps

Notes

Two species of water smartweed, *P. amphibium* L. and *P. coccineum* L., were recently submerged together into one species, *P. amphibium*, and are synonymous.

Water pepper

Water smartweed/knotweed

IV.F.13 Dock (*Rumex* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkansas; edited by Norman Melvin NRCS Wetland Science Institute, Laurel, Maryland, December 2001; photographs by NRCS Plants Data Center, Baton Rouge, Louisiana)

Docks include native and non-native members that are adapted to soil types and water conditions ranging from wet woods and swamps (e.g., swamp dock [*R. verticillatus* L.]) to sandstone and chert dominated soils (e.g., common sorrel [*R. acetosella* L.]). Several species, including bitter dock (*R. obtusifolius*), sour dock (*R. crispus* DC.), and water dock (*R. orbiculatus* Gray) commonly occur on intermediate sites characterized by wet or damp soils. Germination and survival to the seed-producing stage appears to be dependent on the water regime. Many of these species may germinate in moist-soil impoundments if the unit is completely dewatered and the soil dries sufficiently. Normally, however, those species adapted to wetter sites predominate. Germination occurs in early spring on dry to moist sites. Reproduction of annuals is accomplished through seed dispersal, whereas perennials (most species) typically initiate growth from established rootstocks. This species is often associated with other early germinating plant species, such as pigweed and foxtail, that are adapted to drier soils.

Plant value

Seeds are consumed by a variety of waterfowl including mallards and pintails. Although this species normally is sparsely distributed within impoundment, each plant produces a large amount of seed. Seed production as high as 1,500 pounds per acre may occur in small areas.

Frequency of occurrence

Problem: Never a problem

Severe problem: Never a severe problem

Control strategies

Because of its sparse distribution, senescence early in the growing season, and high seed production capabilities, dock is never considered a problem plant. Impoundments with dock typically contain a diversity of other beneficial seed producing plants adapted to similar germination and growing conditions.

Swamp dock

Water dock

IV.F.14 Foxtail (*Setaria* species)

(Text by United States Geological Survey, Moist-Soil Management Advisor, and Jody Pagan, NRCS, Little Rock, Arkanasas; editing and photographs by Norman Melvin, NRCS Wetland Science Institute, Laurel, Maryland, December 2001)

Foxtails (or bristlegrasses) are early successional annual or perennial grasses. This group includes upland and wetland species. In moist-soil areas they commonly occur on dryer sites within moist-soil impoundments. During wet years germination is best following late spring or summer drawdowns. In dry years germination and seed production often is better following early spring drawdowns. Several native and non-native species in this group are valuable seed producers.

Bristlegrass

Foxtail

Plant value

Seeds of foxtails are consumed by many waterfowl species. Seed production may exceed 400 pounds per acre in dense stands. Foxtails do not withstand flooding; stems often form mats above the soil soon after floodup. Therefore, foxtails provide little cover, but may represent an important invertebrate substrate.

Selected species

Giant foxtail (*Setaria magna* Griseb.) is a tall (to 12 ft) native species of fresh or brackish marshes and wetlands in eastern and southern coastal areas.

Japanese bristlegrass or nodding foxtail (*Setaria faberii* Herrm.) grows to about 4 feet and is an introduced species from China. Flowering and producing seed from July through October, it is easily identified by its nodding inflorescence. California and Minnesota both include this species on their state noxious weed list.

Yellow foxtail or yellow bristlegrass (*Setaria pumila* (Poir.) Roemer & J.A. Schultes) is an introduced grass from Eurasia. The scientific name has recently been changed from *S. glauca.*

Frequency of occurrence

Problem: Never a problem (in a moist-soil management area)

Severe problem: Never a severe problem (in a moist-soil management area)

Enhancement

Foxtails are among the best seed producers adapted to dry sites within moist soil impoundments. Thus, they are not considered a problem. Enhancement is difficult because the height of the foxtail normally precludes the use of mowing to reduce competition, and irrigation is not necessary in most years.