Department of Defense
Legacy Resource Management Program

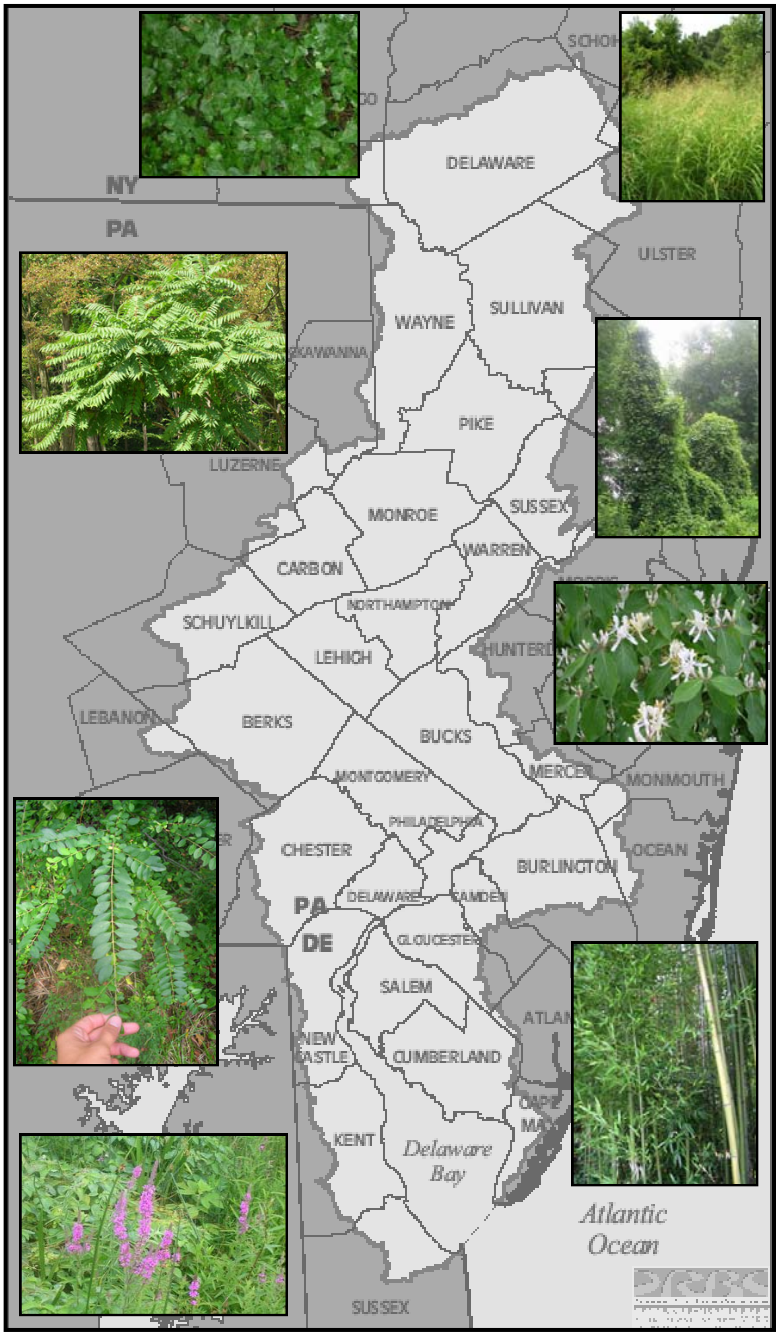
PROJECT NUMBER 08-328

Invasive Species Management at DOD Facilities

Wildlife Habitat Council

June 2009

Invasive Species Guidebook for Department of Defense Installations in the Delaware River Basin: Identification, Control, and Restoration



Adam Gundlach & Susan Reines Robinson

Invasive Species Program Managers, Wildlife Habitat Council

Funded by the Department of Defense Legacy Resource Management Program through Cooperative Agreement W912DY-06-2-0020 between the United States Army Corps of Engineers and Wildlife Habitat Council

ACKNOWLEDGEMENTS

Previous page photographs: Map of Delaware River Basin courtesy of the Delaware River Basin Commission. Photos by Adam Gundlach, Wildlife Habitat Council. Upper left (clockwise): English ivy; johnsongrass, porcelainberry, Amur honeysuckle, golden bamboo, purple loosestrife, privet, tree-of-heaven.

This project was funded by the Department of Defense Legacy Resource Management Program under Cooperative Agreement W912DY-06-2-0020 between the United States Army Corps of Engineers and Wildlife Habitat Council.

Special thanks go out to Commander Scott Merritt, Krista Grigg, Maria Vargas, Laura Muhs, Seth Berry, John Pilcicki and Dorothy Keough for their partnership in coordinating efforts at the pilot sites; Pedro Morales and his colleagues at the DoD Legacy Resource Management Program for support and guidance; Lee Patrick and the crew from Invasive Plant Control, Inc. for their expert herbicide applications and advice; the Delaware River Basin Commission for permission to use the map on the front cover; everyone whose photos we obtained through Invasive.org and the Bugwood Network; and the following people for their generosity in sharing time and information about current invasive species issues and management strategies in the basin: Ben Murdock and the Catskill Regional Invasive Species Partnership; Troy Weldy of The Nature Conservancy; and Art Gover of Penn State University. For their time and input on the original version of this publication (Invasive Species Guidebook for Department of Defense Installations in the Delaware River Basin) , which was used as a basis for this document, we thank LTC William Sames, Dr. Pete Egan, Malcolm Boswell, Richard Claytor, Carolyn Neil, Duane Murphy, Steve Watters and Philip Latasa. Last but certainly not least, we extend thanks to our colleagues at the Wildlife Habitat Council for all of the time and energy they devoted to this publication and the earlier version.

Suggested citation: Gundlach, A.M., and S.B. Reines Robinson. 2009. Invasive Species Guidebook for Department of Defense Installations in the Delaware River Basin: Identification, Control, and Restoration. Wildlife Habitat Council. Silver Spring, Maryland. Prepared for the Department of Defense Legacy Resource Management Program, Project 08-328.

CONTENTS

Introduction

Executive Order (EO) 13112 defines invasive species as "non-native plant, animal, or microbial species that cause, or are likely to cause, economic or ecological harm or harm to human health." Such species have been introduced outside of their natural geographic range by intentional or unintentional human actions (VISC 2005), and have since become naturalized, establishing viable reproducing populations. The problem of invasive species (also referred to as non-native, non-indigenous, exotic, alien, noxious, weed, and pest species) continues to increase in magnitude as new invasive organisms are introduced around the globe, currently established invasive species are dispersed across the landscape – invading approximately 700,000 hectares of wildlife habitat per year in the U.S. (Babbit 1998 in Pimentel et al. 2004) – and further research manifests the negative impacts to native ecosystems that arise from their presence. Researchers calculate that invasive species threaten the existence of somewhere between 35 and 50 percent of endangered and other protected species (Wilcove et al. 1998, Westbrook et al. 2005).

Beyond degradation to ecological communities, invasive species can threaten human health and cause significant economic losses related to decreased productivity in croplands and interference in commerce (e.g. clogged waterways and industrial pipes) (Vitousek et al. 1996 in Mack & Lonsdale 2001). Pimental et al. (2004) estimated that the negative effects and cost of management for invasive species totals more than $120 billion/year in the U.S., a number that will surely increase as new invasive species are introduced and the geographic ranges of existing species expand.

> **"A COUNTRY WORTH DEFENDING IS A COUNTRY WORTH PRESERVING."**
> - Brigadier General Mike Lehnert, Commanding General of Marine Corps Base Camp Pendleton

Reasons for Action

Some may question the importance of invasive species control given other tasks that may seem more relevant to the military mission and current objectives. According to Westbrook et al. (2005), invasive species can impair military operations in four ways. They can:

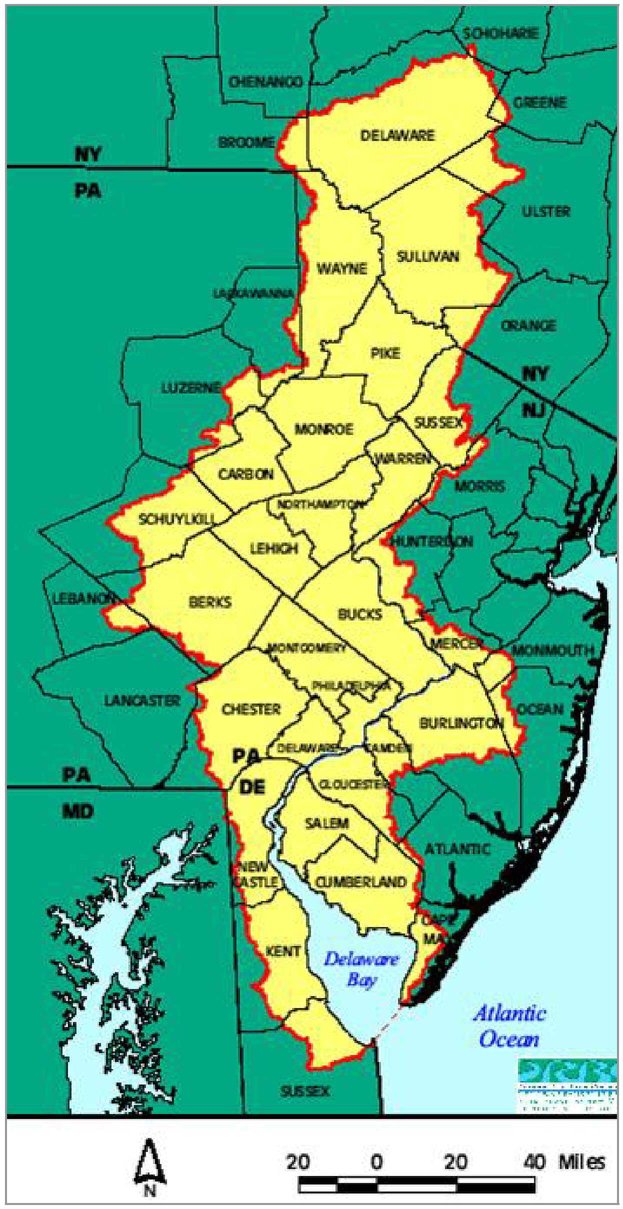
- Eliminate realistic training or testing conditions and limit related activities.
- Divert funding from other natural resource or operational priorities.
- Act as a main cause of habitat destruction and biodiversity loss, further reducing training lands.
- Pose security risks (e.g. creating visual screens) or lead to potentially life threatening situations (e.g. increasing the incidence and intensity of wildfires).

An article by Colonel Robert J. Pratt in a 2004 issue of the U.S. Army's professional journal, Parameters, states, "the homeland is vulnerable to a different type of asymmetric attack, a biological attack from invasive species," and discusses how such biological attacks could pose threats to the nation's economy, food supply, and human health. Though this article likely refers to invasive organisms in all of their forms, invasive plants certainly comprise a large portion of biological invasions across the landscape. The Department of Defense manages 30 million acres of federal land across the United States, and unless invasive species management is approached more aggressively on these lands, the future financial and opportunities costs could be difficult to overcome.

EO 13112 established the duties of federal agencies to prevent the introduction of invasive species, provide for their control, and minimize economic, ecological, and human health impacts associated with invasive species. In pursuit of these goals, the National Invasive Species Council (NISC) – a governing body mandated by EO 13112 and comprised of ten government agencies – created the National Invasive Species Management Plan, which set action items to prevent the spread of invasive species and mitigate their adverse affects (NISC 2001). A key management objective on both national and installation levels must be prevention of invasive species introduction and movement. Though this task is not easily accomplished, it is crucial that the flow of invasive species introductions into natural areas be curbed. Management of existing invasive species on DOD lands must also be approached with greater urgency to protect ecosystem integrity and ensure that adequate, realistic training grounds exist for military maneuvers (Westbrook et al. 2005).

The Delaware River Basin is a crucial area for invasive species control. The basin covers 13,600 square miles across portions of four states: New York, New Jersey, Pennsylvania and Delaware (map below). It includes the metropolitan areas Philadelphia, Trenton and Dover, large military installations such as Fort Dix, and important natural areas like the western slopes of the Catskill Mountains, where the Delaware River's northernmost tributaries originate. Pressure on the once-abundant forest and wetland habitats in the basin is immense. The population grew by almost half a million between 1990 and 2000 and is projected to reach 9 million by 2030. New development accompanying the expanding population is pushing into once-rural areas. In fact, while the watershed population as a whole increases, the population in established urbanized areas such as Philadelphia is declining, suggesting that there is a tendency to develop new land rather than populate urban areas more densely (Delaware River Basin Commission 2008). Thus, it is more important than ever to maintain the health of natural areas as fewer ecosystems remain to support wildlife and provide drinking water, shade, navigable waterways, and countless other resources that are critical to livelihoods and commerce in the basin.

DELAWARE RIVER BASIN



Source: Delaware River Basin Commission

Notes about this Publication

This publication was developed through a cooperative agreement (W912DY-06-2-0020) between the Wildlife Habitat Council (WHC) and the U.S. Army Corps of Engineers (USACE), working under the Department of Defense Legacy Resource Management Program. Many documents regarding invasive species have previously been produced and were instrumental in the development of this guidebook. A main goal of this document is to provide DOD installations and personnel with pertinent information to increase the efficiency and effectiveness of management activities while minimizing interference to their primary duties and military mission.

The first part of this guidebook covers the most detrimental invasive plant species encountered in or threatening the Delaware River Basin, with pictures and descriptions of each species to aid identification and appropriate control methods. Plants are only one type of invasive organism that threatens the basin, but this document focuses on plants specifically because that is the type of invasive organism most land managers have the capability to address. Control of other invasive organisms such as fish, mammals and algae is also critically important; government agencies that deal with natural resources (such as a local office of the U.S. Fish & Wildlife Service) are the best resources for developing programs to control them.

The second part of this guidebook discusses strategies and management activities to prevent recurrence of problem invasive species. This section also gives recommendations for returning management areas to historical native plant communities. The third part discusses the successes and obstacles encountered while coordinating and implementing invasive species control at pilot sites.

Section I – A:
Invasive Plant Species Identification

This section covers invasive plant species that pose the greatest ecological threat in the Delaware River Basin. The plants described were chosen from state and federal noxious weed lists as well as lists developed by conservation organizations and invasive species working groups throughout the basin. The species included do not entail all invasive species present in the basin but do represent some of the most troublesome species. The descriptions provide identification information and control methods for each species. Learning how to identify pest species and where they are most likely to occur will facilitate early detection of their presence and increase the likelihood of successful control. Early detection and rapid response can make the difference between a manageable population and one that is not technically or financially feasible to control (NISC 2001). Regardless of the species, invasive plant control requires consistent long-term monitoring and management to ensure success, as most species develop an abundant soil seed bank, have extensive perennial root systems, or a combination of traits that allows them to persist at a site for extended periods once established.

Characteristics typical of many invasive species (National Research Council 2002):

- Long flowering/fruiting period, which increases seed production and dispersal.
- Ability to reach reproductive maturity quickly.
- Seeds remain viable (or dormant) for extended periods, allowing populations to wait for favorable conditions before germinating.
- Efficiently use light, water, and nutrients in the environment.
- Well-developed root system.

Each species description includes a brief summary of control methods that have proven effective in management, as well as methods that have not. More detailed discussions of specific control methods can be found in Section I – Part B: Management Techniques.

Additionally, the U.S. Army Corps of Engineers (USACE), Engineer Research and Development Center (ERDC), has developed two powerful tools to assist with identification and management of terrestrial and aquatic plants. The Noxious and Nuisance Plant Management Information System (PMIS) and the Aquatic Plant Information System (APIS) provide detailed information regarding a wide range of plant species and control techniques. Both the PMIS and APIS tools are updated on a regular basis. CD-ROM versions can be requested through the respective websites (linked above).

INVASIVE HERBACEOUS PLANTS

COMMON NAME	SCIENTIFIC NAME	PAGE
Goutweed	*Aegopodium podagraria*	3
Garlic mustard	*Alliaria petiolata*	4
Common (lesser) burdock	*Arctium minus*	6
Giant reed	*Arundo donax*	7
Musk thistle	*Carduus nutans*	9
Spotted knapweed	*Centaurea biebersteinii*	11
Canada thistle	*Cirsium arvense*	13
Crown vetch	*Coronilla varia*	15
Leafy spurge	*Euphorbia esula*	16
Ground ivy	*Glechoma hederacea*	18
Giant hogweed	*Heracleum mantegazzianum*	19
Cogongrass	*Imperata cylindrica*	21
Chinese lespedeza	*Lespedeza cuneata*	23
Purple loosestrife	*Lythrum salicaria*	25
Japanese stiltgrass	*Microstegium vimineum*	27
Chinese silvergrass	*Miscanthus sinensis*	29
Wavyleaf basketgrass	*Oplismenus hirtellus* ssp. *undulatifolius*	30
Japanese spurge	*Pachysandra terminalis*	32
Reed canary grass	*Phalaris arundinacea*	33
Common reed	*Phragmites australis*	35
Golden bamboo	*Phyllostachys aurea*	37
Japanese knotweed	*Polygonum cuspidatum*	39
Lesser celandine	*Ranunculus ficaria*	41
Japanese bristlegrass	*Setaria faberi*	42
Johnsongrass	*Sorghum halepense*	43

Adapted from PCA Alien Plant Working Group – Mid-Atlantic List

Goutweed (*Aegopodium podagraria*)

Goutweed (variegated form) foliage. Photo by David Schimpf, Department of Biology, University of Minnesota – Duluth. Courtesy of PCA Weeds Gone Wild.

Description & Biology – Goutweed, also called bishop's-weed or snow-on-the-mountain, is a perennial herbaceous plant that is native to Europe and Asia. Goutweed is a low-growing plant that produces leaves mainly from basal leaf stalks attached to underground stems, called rhizomes. The leaves are green and comprised of three groups of three leaflets (triternate), which are toothed and sometimes irregularly lobed. A common variegated form of the plant used in landscaping has bluish-green to green leaflets with creamy white margins. Small white flowers bloom in mid-summer and are arranged in numerous clusters (called compound umbels) produced at the end of flower stalks, which can reach three feet tall. Small seeds, similar to those produced by carrots, ripen in late summer, but are not a significant source of reproduction, as they require cold stratification to germinate and open, disturbed sites in full sun to become established. Once established, goutweed mainly reproduces vegetatively through spread of rhizomes – long, white, branching underground stems – that allow it to invade shaded environs. From the rhizomes, it is capable of forming dense patches in the ground layer that can exclude most native vegetation. The primary dispersal vector for goutweed is movement and planting by humans.

Goutweed foliage with flower stalks. Photo by David Schimpf, Department of Biology, University of Minnesota – Duluth. Courtesy of PCA Weeds Gone Wild.

Goutweed is an aggressive invader that grows best in well-drained, moist soils in partial shade, though it is capable of growing in full shade. It tolerates a variety of soil conditions and typically is found in disturbed habitats near old flowerbeds, from which it can invade surrounding natural areas, including closed-canopy forests. Infestations of goutweed greatly reduce species diversity in the ground layer and prevent regeneration of native tree species.

Control – Hand removal of goutweed is generally not effective unless performed routinely and carefully to remove all portions of the rhizome, which if left in the soil, can sprout to form new plants. Discard rhizomes properly and avoid spreading contaminated compost or yard waste. Large infestations can be controlled using systemic herbicides, such as glyphosate (e.g. Accord®, Roundup Pro®) or triclopyr (e.g. Garlon®), which are transported to the roots, effectively killing the entire plant. Contact herbicides are not as effective, as goutweed develops new leaves after being defoliated. No biological control agents are available for goutweed.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and U.S. Forest Service Weed of the Week.

Garlic mustard (*Alliaria petiolata*)

Description & Biology – Garlic mustard is a cool-season, biennial herb of the mustard family (Brassicaceae) introduced to North America in the mid-19th century by European settlers. It is occurs in most northeastern and Midwestern states and several western states. During its first season of growth, garlic mustard seeds germinate in April or May and produce a rosette of low-growing, round or kidney-shaped, leaves that remain green through the winter. The rosette stage may be confused with native rosette-forming species, such as violets (*Viola* spp.), white avens (*Geum canadense*), and toothworts (*Cardamine* spp.) During the following spring, the adult plants produce flowering stems – a process called bolting – that range in height from two to four feet. Leaves originating from the flower stem are alternate, coarsely toothed, and triangular or heart-shaped. Young leaves produce a garlic odor when crushed. In April or May, flowers consisting of four white petals form in clusters at the end of stems. Garlic mustard flowers are cross-pollinated by insects, but are also capable of self-fertilization depending on the conditions. A single plant may produce more than one thousand shiny, black seeds, which develop inside green, tubular pods (called siliques) that become tan and papery as they mature. Plants that develop from self-fertilized seeds are genetic clones of the parent, allowing a single plant to infest an area. Seeds are generally only dispersed in the immediate vicinity of the parent plant; however, humans, wildlife, and water aid long distance dispersal. Most second-year plants die off by the end of June, leaving behind only dead flower stalks with dry siliques. Garlic mustard reproduces solely by seed, with each plant capable of producing hundreds to thousands of seeds that are dispersed in the vicinity of the parent plant. The seeds may remain viable for five years or more.

Garlic mustard grows in a variety of moist to dry habitats, frequently observed in forests, floodplains, forest edges, hedgerows, and roadsides. It poses the greatest threat to moist forest and riparian plant communities, as its shade tolerance, early cool-season growth, and

Garlic mustard first-year rosette. Photo by Chris Evans, River to River CWMA, Bugwood.org

Garlic mustard flowers and leaves. Photo by Chris Evans, River to River CWMA, Bugwood.org

A flowering second-year garlic mustard plant. Photo by Chris Evans, River to River CWMA, Bugwood.org

prolific spread can displace many native understory herbs. It is often associated with calcareous soils and does not tolerate acidic soils. Garlic mustard invasions result in a significant decline in native plant diversity, which in turn impacts invertebrate populations. Several butterfly species, particularly the rare West Virginia white (*Pieris virginiensis*), can be significantly impacted when garlic mustard populations displace their native host plants.

The butterflies lay their eggs on garlic mustard in the absence of native host plants, but most of these larvae die before developing into adults. (While some research suggests that the butterflies may be adapting to the invasive plants, evolution may not be quick enough to save sensitive and rare species like the West Virginia white.) Garlic mustard also produces allelopathic chemicals in the soil that discourage growth of native plants by impacting the symbiotic mycorrhizal fungi that they rely on.

NOTE: Garlic mustard may grow with native species that are similar in appearance, such as toothworts (*Dentari*a spp.), sweet cicely (*Osmorhiza claytonii*), and early saxifrage (*Saxifraga virginica*).

Garlic mustard seedpods, called siliques. Photo by Chris Evans, River to River CWMA, Bugwood.org

Control – Various control techniques may be used for garlic mustard depending on the size of the infestation, the location, and the resources available. A successful control program will prevent seed production for successive years until the seedbank is exhausted. A single plant overlooked can form a new infestation. Though labor intensive, small patches can be hand pulled prior to seed set, removing as much of the root as possible to prevent any resprouts from root fragments. Plants can also be cut at ground-level during the flowering stage to prevent seed production. Cutting plants before the flowering stage may allow them to develop new flower stalks prior to senescence. All plant material should be bagged and removed from the site. For extensive infestations where damage to non-target vegetation is not a concern, systemic herbicides (glyphosate, triclopyr) may be applied prior to seed set. Herbicides can be applied year-round to first-year rosettes as long as temperatures are sufficiently warm (consult herbicide label). Burning for successive years has shown mixed results, and may only serve to increase an infestation. Several biological control agents (*Ceutorhynchus* spp. and *Phyllotreta ochripes*) are being studied in Europe to determine their suitability for release in the United States.

Compiled from the PCA Alien Plant Working Group Fact Sheet, Southeast Exotic Pest Plant Council Plant Manual, and The Nature Conservancy Element Stewardship Abstract, and the following article:

Keeler, M.S. & F.S. Chew. 2008. Escaping an evolutionary trap: preference and performance of a native insect on an exotic invasive host. Oecologia 156: 559-568.

Common (lesser) burdock (*Arctium minus*)

Description & Biology – Common burdock is known to harbor pathogens such as powdery mildew and root rot and thus has the potential to severely impact both natural areas and agricultural sites. The species is introduced from Europe and has deadly impacts on North America's native wildlife, particularly hummingbirds, which can become ensnared in the extremely prickly burs. The burs, sometimes called "hitchhikers," are thought to have inspired the invention of Velcro.

Old fields, fencerows, and other places where there has been past disturbance are common colonization sites. Extensive patches can form in natural areas where there has been soil disturbance. In the first year of growth, leaves emerge in a rosette. Young leaves are egg-shaped, becoming heart-shaped with age. The basal leaves can get as large as 18 inches by 14 inches, and they have hairy undersides. Leaf margins become markedly wavy and toothed as the leaves mature, and a reddish midvein becomes apparent. If conditions are good, in the second year the plant produces a stem (up to 5 feet in height, hollow, hairy, ridged, stem leaves are alternate) with purple (occasionally pink or white) clusters of flowers. In some cases plants may not flower until the fourth year or later. The flowers dry into prickly burs, which cling to passing animals for effective distribution of the thousands of seeds produced by each plant. Each plant has a long, fleshy taproot.

Control – Digging is not advised, as soil disturbance promotes germination of the seed bank, and the long taproots are difficult to extract. Fortunately, repeated cutting to ground level prior to flowering is effective. If any flower parts or burs are present, they should be bagged and discarded. Monitoring after cutting is essential to remove regrowth. Glyphosate also effectively controls burdock.

Compiled from the Virginia Tech Weed Identification Guide, Colorado State Parks Best Management Practices, University of Wisconsin Weed Science page, the Fletcher Wildlife Garden fact sheet, and Nealen & Nealen 2000.

(top) Heart-shaped basal leaves. (middle) Flowers with developing burs beneath. (above) Flowering plant. Photos by Richard Old, XID Services, Inc., Bugwood.org

Giant reed (*Arundo donax*)

Description & Biology – Giant reed is a perennial grass native to Asia and the Mediterranean region that produces large stems (called culms) resembling corn, which reach more than 20 feet in height. Leaves are arranged alternately along the culms, are one to two inches wide, 12 inches long, and taper to a point. Giant reed produces dense root masses that send roots deep into the soil and form persistent, drought-resistant rhizomes. Flowers are borne in terminal plumes of whorled spikelets that may grow to as much as three feet in length. Reproduction of giant reed is mainly vegetative through rhizomes and stem fragments, which root and sprout readily when transported to suitable locations. Further research is needed regarding the importance of sexual reproduction for giant reed, but it is believed that most seeds produced by plants in the United States are not viable.

(top) Giant Reed infestation with flower plumes. Photo by Chuck Bargeron, University of Georgia, Bugwood.org. (above) Giant Reed stalk and leaves. Photo by James H. Miller, USDA Forest Service, Bugwood.org

Giant reed tolerates a variety of environmental conditions and soil types ranging from high salinity, to clay, to loose sandy soils. It grows best in well-drained soils with abundant moisture, often becoming established along ditches, streams, riverbanks, and other similar areas. Mature plants are able to tolerate extended drought because of hearty rhizomes and a deep-penetrating root system. Once established in an area, its high rate of growth and vegetative reproduction allow it to form dense monotypic stands that crowd out native vegetation. Root and stem fragments can be transported downstream to form new infestations. This aggressive growth and spread decreases wildlife habitat, increases the risk of fire, and causes problems with flood control.

Control – Small populations can be removed via manual or mechanical methods. Hand removal by cutting and digging up rootstock can be effective, though labor-intensive. Mowing may control the spread of populations, but is often not effective due to the large amount of biomass produced by giant reed, its persistent rhizomes, and its ability to resprout from stem and root fragments. Repeated mowing for consecutive years may exhaust energy reserves in the root system. Mowing and cutting machinery often cannot access areas where giant grows due to saturated or easily compacted soils. For extensive stands of giant reed, systemic herbicide, such as glyphosate (Rodeo®), will effectively kill both above and belowground growth. It can be applied as cut-stump or foliar application, and may be combined with a prescribed burn to remove the thatch layer. Herbicide applications should occur after the flowers emerge in August and September when energy reserves are lowest. Be sure to use appropriately labeled herbicides in wetland environments.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, Plant Invaders of Mid-Atlantic Natural Areas, and The Nature Conservancy Element Stewardship Abstract.

Musk thistle (*Carduus nutans*)

Description & Biology – Musk thistle, also called nodding thistle because of its large, drooping flower head, is a biennial herb native to Western Europe. It generally develops over the course of two growing seasons, but may germinate and produce flowers in a single season in warmer climates. The typical biennial life cycle begins in July with seed germination and development of seedlings. The seedlings form a low-growing rosette of coarsely lobed leaves that can reach a diameter of four feet. Leaves are dark green with white along their margin and a light green midrib. Plants overwinter in the rosette stage and then bolt (produce flowering stems) the following March. Stems with numerous branches are produced during the bolting stage and mature plants may grow to six feet tall. The showy pink or purple flower heads emerge from May to August and droop to the side as they mature. The number of flower heads produced per plant varies depending on site conditions, ranging from one flower head to near 60. Each flower head is capable of producing more than one thousand seeds, which are wind-dispersed and remain viable for ten years or more.

Musk thistle rosette. Photo by Steve Dewey, Utah State University, Bugwood.org

Musk thistle often invades meadows, prairies, pastures, rights-of-way, and other disturbed open areas. It typically is not a threat to healthy natural areas with diverse assemblages of native plants. It is unpalatable to livestock and wildlife, giving it a competitive advantage over native vegetation that allows it to invade pastures and meadows. As other vegetation is selectively grazed, competition for nutrients, light, and water is reduced and new areas of disturbance are created for musk thistle colonization.

Control – Small populations can be hand pulled prior to seed set, bagging flower heads to prevent dispersal of seeds. Avoid excessive soil disturbance to prevent germination of existing seeds in the soil. Several herbicides, including glyphosate (e.g. Accord, Roundup Pro), triclopyr (e.g. Garlon), and Chlorpyralid (e.g. Transline®), are effective for controlling larger populations of musk thistle. Foliar sprays should be applied to during the

(above left) Flowering musk thistle plant. Photo by Norman E. Rees, USDA Agricultural Research Service, Bugwood.org. (above right) Flower heads. Photo by Ricky Layson, Ricky Layson Photography, Bugwood.org.

rosette stage or before flower production. Triclopyr is selective for broad leaf species and is a good choice for areas with native grasses. Multiple biological control agents have been released to control musk thistle, including several weevil species (*Rhinocyllus conicus*, *Trichosirocalus horridus*, *Ceutorhynchos trimaculatus*), a fly (*Cheilosia corydon*), and a beetle (*Psylliodes chalcomera*). Effectiveness of biocontrol agents on musk thistle has varied, with *R. conicus* and *T. horridus* weevils exhibiting the greatest impacts; however, *R. conicus* has been observed to feed on native thistle species as well.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and Southeast Exotic Pest Plant Council Plant Manual.

Spotted knapweed (*Centaurea biebersteinii*)

Description & Biology – A native of Eurasia, spotted knapweed was introduced to the United States in the late 1800's as a contaminant in alfalfa and discarded ship ballast. It is a biennial or short-lived perennial plant named for its black tipped flower bracts that create a spotted appearance. In North America, spotted knapweed plants may live from three to ten years. A basal rosette of eight-inch long by two-inch wide leaves develops in the first year. Leaves are borne on short stalks and have one to two deep lobes on either side of the mid-vein. Flowering stems two to four feet tall develop in subsequent years from buds on the root crown. Stems are slightly or densely hairy (pubescent) with leaves alternating along the stem and decreasing in size toward the stem tip. Plants produce abundant flower heads at the end of stems. The purple or pink flowers bloom from June to October, and the oval-shaped flower heads generally remain on the plant after blooming. Reproduction is almost exclusively by seed, with each plant capable of producing thousands of seeds that may remain viable in the soil for five to eight years. Spotted knapweed also produces rhizomes from the root crown that form rosettes adjacent to the parent plant.

(top) Spotted knapweed plant. Photo by Michael Shephard, USDA Forest Service, Bugwood.org. (middle) Spotted knapweed flower head and rosette (bottom). Photos by Steve Dewey, Utah State University, Bugwood.org

Spotted knapweed is most commonly associated with habitats that receive full sun and have loose, well-drained soils, but it is adapted to a range of habitats and soil types. It typically invades disturbed sites, roadsides, and fields in the eastern U.S. Its deep taproot provides it with access to water during times of drought, giving it a competitive advantage over other plant species. Spotted knapweed roots exude allelopathic chemicals into the soil that inhibit germination and growth of competing plant species. This feature combined with its prolific seed production allows it to quickly invade suitable habitats.

Control – Manual pulling can be performed prior to seed set, but the entire taproot and root crown must be removed to prevent sprouting. Gloves should be used to prevent skin irritation. A variety of biocontrol agents, including seed head flies (*Urophora affinis and U. quadrifaciata*), moths (*Agapeta zoegana, Pelochrista medullana, Pterolonche inspersa*), and weevils have been released in the United States to control spotted knapweed. Five biocontrol agents have become established in eastern states. Large infestations can be treated with various herbicides, including 2,4-D, clopyralid (Transline®), and picloram (Tordon K®). Picloram is a persistent herbicide that can achieve control for three to five years; however, it can also be a potential groundwater contaminant in locations with permeable soils or a shallow water table. Herbicide applications will likely need to be repeated for multiple years or combined with other control methods due to continued germination of seeds from the seedbank. Planting native grasses and forbs can suppress reproduction of knapweed following control activities. Long-term grazing by sheep and goats has also shown some success.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and U.S. Forest Service Weed of the Week.

Canada thistle (*Cirsium arvense*)

Description & Biology – Canada thistle is a highly invasive perennial herbaceous plant introduced to North America in the 1600's from Eurasia. It grows from one to four feet tall with spiny, irregularly lobed leaves that alternate along branched, slightly hairy stems. Fragrant flowers are produced from June through October and can vary in color from purple to occasional white. Canada thistle is a dioecious plant; male and female flowers develop on separate plants as rounded clusters. Individual plants can produce thousands of small, brown seeds with bristly plumes that are easily wind-dispersed. Seeds generally germinate the following year but can remain dormant in the soil for more than 20 years before germinating. Canada thistle reproduces mainly by vegetative means and develops a deep, fibrous taproot, which can extend six feet deep. Creeping lateral roots arise from the taproot and produce numerous shoots from adventitious root buds. Root fragments separated from the parent plant can sprout and develop into new plants. Canada thistle can be distinguished from other thistle species by its creeping root system and clonal growth.

(top) Canada thistle flower heads. (above) Canada thistle leaves and stem. Photos by Mary Ellen (Mel) Harte, Bugwood.org.

Canada thistle invades a variety of open habitats such as fields, meadows, barrens, savannas, and prairies. Although it grows best in disturbed upland habitat, it also colonizes locations that experience periodic inundation such as stream banks and wet prairies. It can be found growing in soils ranging from gravel to clay and is a major pest of agricultural crops. In natural areas, Canada thistle infestations displace native plants and reduce overall biodiversity.

(top) Canada thistle flower heads. (above) Canada thistle leaves and stem. Photos by Mary Ellen (Mel) Harte, Bugwood.org.

Control – Canada thistle is easiest to control in early stages of invasion. A combination of control methods is often most effective at achieving control. Small populations can be cut by hand and larger populations can be mowed. Both techniques should be performed prior to seed set and must be repeated multiple times per growing season for several years before the root reserves are exhausted. Dense stands of Canada thistle are best treated with systemic herbicides (glyphosate, triclopyr, clopyralid). The best application times are late summer and early fall when translocation to the roots is greatest. Efficacy can be increased by cutting plants in late July and spraying the resprouts in late August. Multiple biocontrol agents have been released throughout North America to control Canada thistle, though their impact has been limited and further research is needed. Seeds are generally not a major source of spread, but their long viability and the hardiness of the root system necessitates repeated control efforts for many years.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, The Nature Conservancy Element Stewardship Abstract, and Van Driesche et al. 2002.

Crown vetch (*Coronilla varia*)

Crown vetch flower clusters. Photo Dave Powell, USDA Forest Service, Bugwood.org

Description & Biology – Crown vetch is a low-growing perennial legume in the pea family (Fabaceae) that is native to portions of Europe, southwestern Asia, and northern Africa. Its stems can reach six feet long, with a creeping growth habit that forms dense clumps. Compound leaves arising from the stem extend up to six inches long and are comprised of 10 to 25 smaller leaflets. Crown vetch blooms from May through August, producing clusters (called umbels) of pink or light purple flowers borne on a short flower stalk. Narrow, multi-segmented seed pods develop, with each segment containing a seed that may remain viable in the soil for several years. It spreads vegetatively by a creeping rhizome system.

Because of its fast growth and development of an extensive root system, crown vetch has been planted extensively throughout the United States for erosion control and bank stabilization along roadsides and other areas of disturbance. It tolerates a variety of conditions but is intolerant of shade and extreme cold. Generally, it is found along roadsides, fields, and other open, well-drained sites. Mature stands of crown vetch are a major problem for prairies, meadows, and grasslands, as their dense growth blankets the landscape and prevents regeneration of native plants.

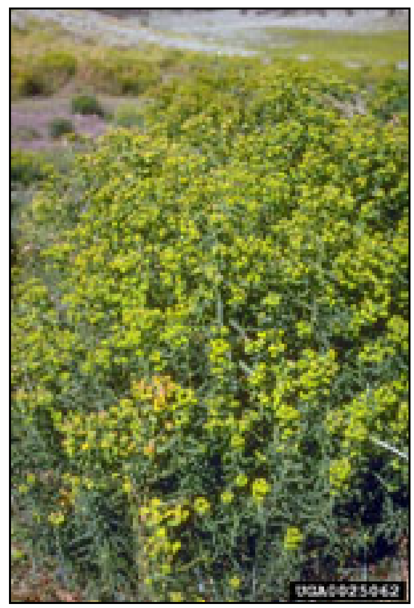
Control – Crown vetch may be confused with native vetch (*Vicia*) species, so careful identification is critical. Small patches of crown vetch can be controlled by repeated pulling. The entire plant, including as much of the root system as possible, should be removed to discourage resprouts. Root fragments left in soil can sprout to form new plants. Larger patches can be repeatedly mowed during the flower bud stage for several consecutive growing seasons to prevent seed production and the spread of crown vetch populations. If repeated regularly, mowing will eventually kill the root system. Herbicide applications are the most effective method to control large infestations. Systemic herbicides (glyphosate, triclopyr, clopyralid) will kill the root system, though follow up spot treatments may be necessary.

Crown vetch compound leaves and flowers arranged along the stem. Photo Dave Powell, USDA Forest Service, Bugwood.org.

Compiled from the Southeast Exotic Pest Plant Council Plant Manual, The Nature Conservancy Element Stewardship Abstract, and U.S. Forest Service Weed of the Week.

Leafy spurge (*Euphorbia esula*)

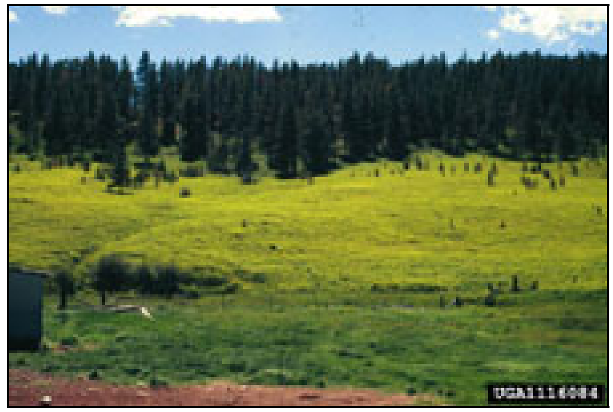
Description & Biology – Leafy spurge is an aggressive perennial herb that produces tough, smooth, multi-branched stems, which can grow to more than three feet in height. When broken, the stems of leafy spurge exude a milky white sap that can cause skin irritation in humans and, if ingested in large quantities, can cause death in livestock. Leaves of leafy spurge are small, slender, and slightly frosted in appearance. Its tiny flowers develop in greenish yellow structures that are surrounded by showy, yellow bracts, which make it appear as though leafy spurge has bright yellow flowers. The yellow bracts open in late May or June prior to the opening of flowers in late June. Grayish brown seeds are produced in three-parted seedpods. The pods open explosively when mature, dispersing seeds up to 15 feet from the parent plant. Seeds readily germinate and remain viable for seven years or more. Leafy spurge forms a complex root system that can extend more than 15 feet below the soil surface. Lateral roots spread rapidly and have numerous adventitious buds that send up shoots to form new plants, allowing leafy spurge to form dense patches in a short time span. It is also capable of regenerating from root fragments.



(top) Leafy spurge flowers and upper leaves. Photo by USDA APHIS PPQ Archives, Bugwood.org. (above) Leafy spurge plants. Photo by Norman E. Rees, USDA Agricultural Research Service, Bugwood.org.

The aggressive growth of leafy spurge displaces native vegetation by shading seedlings and dominating nutrients and water. Allelopathic compounds produced by leafy spurge also prevent growth of competing vegetation. It grows best in arid conditions but tolerates a variety of soil and moisture combinations. Generally, it invades areas of disturbance, such as prairies, pastures, abandoned fields, and roadsides.

Control – Controlling leafy spurge infestations is difficult and requires dedicated management for several years due to the persistent root system – even small root fragments can form new plants – and longevity of seeds in the soil. For these reasons, manual control is not effective. Application of systemic herbicides, including picloram, dicamba, 2,4-D, and glyphosate, have proven to be the most effective method for long-term control. Applications should take place in June prior to production of seeds or in September when nutrients are being translocated to the root system. Picloram is often most effective because of residual soil activity that persists for several years, but it is not ideal in all situations, as its soil persistence can also cause water contamination in locations with a shallow water table. Read all labeling thoroughly. Six species of insects collected from Europe have been released in the United States as biological control agents. Their impacts are not as immediate as herbicide treatments, but once populations become established and reproduce for several years, their impacts on leafy spurge populations are impressive.



Rangeland infestation of leafy spurge. Photo by Norman E. Rees, USDA Agricultural Research Service, Bugwood.org.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, The Nature Conservancy Element Stewardship Abstract, and Van Driesche et al. 2002.

Ground ivy (*Glechoma hederacea*)

Description & Biology – Ground ivy, also called cat's foot, creeping Charlie, field balm, and gill-over-the-ground, is a low-growing perennial herb native to Europe. Like other members of the mint family (Lamiaceae), ground ivy has square stems that grow more than a foot in length, trailing across the ground and rooting at the nodes to form dense mats, which crowd out other vegetation. The oppositely arranged, kidney-shaped leaves are deep green in coloration with bluntly toothed margins. From March through May, numerous whorled flowers are borne on short flower stems. The tube-shaped flowers are violet to bluish in coloration and produce a pod containing four nutlets. Ground ivy develops shallow, fibrous roots that arise from the base of stems and leaf nodes, allowing the plant to spread vegetatively once established.

Ground ivy typically invades open woods, lawns, disturbed areas, forest edges, and any other areas with damp, fertile soil. It is intolerant of acidic soils and salinity.

(top) Flowering ground ivy. (above) Creeping growth along the ground. Photos by Adam Gundlach, Wildlife Habitat Council

Control – Ground ivy can be difficult to control because of its ability to regenerate from root and stolon fragments and dormant seeds. Manual removal should be performed when soil is damp and should attempt to remove all stem and root fragments from the soil. Applications of herbicides such as glyphosate, 2,4-D, and dicamba are effective for extensive infestations. Applications should be performed in early spring when ground ivy is flowering, but prior to seed set. Spot applications can be repeated in the following years to eliminate plants that develop from the seedbank. A newly discovered rust fungus (*Puccinia glechomatis*) may potentially be used as a biocontrol agent, as severe infections of the fungus can kill entire sections of the plant.

Compiled from the U.S. Forest Service Weed of the Week.

Giant hogweed (*Heracleum mantegazzianum*)

Description & Biology – Giant hogweed is a priority for control because of its public health impacts. This species contains a sap that, when combined with moisture (e.g. perspiration) and sunlight, causes severe skin blistering. Skin can be contaminated with sap as a result of brushing against the bristles or touching broken stem or leaf parts. Blisters appear 15 to 20 hours after contact. Contact with eyes can result in severe impacts to vision. If contact occurs, skin should be covered to block sunlight and immediately washed thoroughly with soap and water. Steroid cream prescribed by a doctor may be necessary for severe reactions.

(top) Giant hogweed leaves and flowers. Photo by Donna R. Ellis, University of Connecticut, Bugwood.org. (middle) Standing stems in winter. Photo by Randy Westbrooks, U.S. Geological Survey, Bugwood.org. (above) Close-up of flowers. Photo by Thomas B. Denholm, New Jersey Department of Agriculture, United States.

Giant hogweed currently exists in scattered areas of the Delaware River Basin, notably in Carbon County, PA. Yards and gardens are common sites of occurrence, possibly because the species has moved within gardening circles as a curiosity. The plant's height (can reach 15 or more feet) and enormous flower clusters (up to 2.5 feet in diameter) make it an unfortunate source of horticultural interest. In addition to the health hazard it poses to humans, giant hogweed degrades ecosystems by outcompeting native plants and creating large monotypic stands that die back in winter, leaving bare patches of ground that are vulnerable to erosion.

In addition to its height (native look-alikes generally do not reach more than 9 feet), giant hogweed can be identified by purple blotches and coarse white hairs on the stem. The native cow parsnip (*Heracleum lanatum*) has softer fuzzier hairs, smaller flower clusters of flowers, and lacks blotches on the stem (though the stem may have a purplish cast). Poison hemlock (*Conium maculatum*) has purple blotches on the stem but lacks hairs. It is important to note that some look-alikes, such as the poisonous hemlock, are also extremely dangerous. The New York State Department of Environmental Conservation offers help identifying giant hogweed at lflands@gw.dec.state.ny.us and (518) 402-9425.

Control – Due to potential for injury and difficulty of control (mechanical control alone is ineffective; many herbicides are ineffective), it is strongly recommended that anyone with a suspected giant hogweed specimen or population contact the state government for assistance:

- New York: The New York State Department of Agriculture & Markets has a giant hogweed hotline at (800) 554-4501 extension 72087.
- Pennsylvania: The Pennsylvania Department of Agriculture has a hotline at (877) 464-9333.
- Delaware: The plant is not yet known to occur in Delaware, and the Delaware Department of Agriculture requests that all potential sightings be reported at 302-698-4500.
- Maryland: The Maryland Department of Agriculture has a survey form that can be filled out to report suspected occurrences. The department can be reached at (410) 841-5700.
- New Jersey: The New Jersey Department of Agriculture is collecting reports of sightings at (609) 984-2262.

Compiled from: New York State Department of Health Health Advice site, New York State Department of Environmental Conservation Nuisance & Invasive Species site, Pennsylvania Department of Agriculture 2008 eradication and control report, Pennsylvania Department of Agriculture giant hogweed brochure and US Forest Service Weed of the Week.

Cogongrass (*Imperata cylindrica)*

(top) Close up of cogongrass leaf with characteristic white midrib. Photo by Mark Atwater, Weed Control Unlimited, Inc., Bugwood.org (above) Rhizome extending laterally from cogongrass roots. Photo by James H. Miller, USDA Forest Service, Bugwood.org.

Description & Biology – A large perennial grass native to Southeast Asia, cogongrass produces substantial rhizomes and grows to four feet in height. Although cogongrass is more suited to the southern United States, it has been known to survive in Virginia, Maryland, and West Virginia. Its sharply pointed leaves are about one-inch wide with finely toothed margins and a conspicuous white midrib that is generally slightly off-center. The upper surface of the leaf is hairy near the base, while the undersurface is generally hairless. Small flowers are arranged in silvery plumes (called panicles) that can reach nearly one foot long and more than an inch wide. A single plant is capable of producing thousands of tiny seeds that can be dispersed long distances by wind. Vegetative reproduction is made possible through hearty rhizomes that can remain dormant for long periods before sprouting. The sharp-tipped, branching rhizomes are pale and segmented, can reach several feet deep (though not typical), and may grow up to ten feet per year from established plants.

Cogongrass is a robust species that can survive in a variety of environmental conditions including shade, high salinity, and drought. It grows in habitats ranging from dry uplands that receive full sun to shaded mesic sites, often invading roadsides, fields, sand dunes, grasslands, swamps, riparian areas, and scrub habitat. Once established in an area, it quickly spreads to form a dense layer of leaves that prevents growth of other vegetation, displacing native plant species and the organisms that rely on them. Extensive infestations can alter fire regimes through increased fuel loads, resulting in fires of greater frequency and intensity that kill native plants.

Control – Cogongrass management requires an integrated approach that combines mechanical removal, herbicide applications, and cultural treatments to prevent recurrence of invasions. Young infestations are easier to control than mature, established stands. If a dense thatch layer exists, it should be removed through mowing or burning to promote new growth and improve conditions for herbicide application. Stands of cogongrass produce intense fires that burn hot and fast, so thorough planning and preparation should take place prior to initiating a prescribed burn. The herbicides glyphosate (e.g. Accord, Roundup Pro) and imazapyr (e.g. Arsenal®) – alone or in combination – have proven effective in controlling cogongrass. The best application time is in the fall prior to the first frost. Imazapyr has high residual soil activity, which could allow it to leach into groundwater or damage surrounding vegetation that has roots extending into the treatment area. After the majority of a cogongrass infestation has been controlled, the area should be planted with fast-growing native species to discourage reinvasions and reduce erosion. Spot treatments will be necessary for several years following initial treatments to control new plants that arise from rhizomes and the seed bank. Rhizome fragments and seeds of cogongrass can be transported to new sites by vehicles, heavy machinery, and contaminated fill dirt. All machinery used in control efforts should be thoroughly cleaned prior to leaving the treatment area to prevent transport of propagules to a new location.

(above left) Cogongrass infestation. (above right) Feathery plumes of cogongrass seedheads. Photos by Chris Evans, River to River CWMA, Bugwood.org.

Compiled from the PCA Alien Plant Working Group Fact Sheet and Van Driesche et al. 2002.

Chinese lespedeza (*Lespedeza cuneata*)

Description & Biology – Chinese lespedeza, also called sericea lespedeza and Chinese bush clover, is a perennial, warm-season, nitrogen-fixing herb native to eastern portions of Asia. Its erect, semi-woody stems grow from three to five feet in height with alternate leaves arising from the stems. Leaves are composed of three small, slender leaflets that have a tiny spine at their apex. Dense, flattened hairs cover the leaflets and give some leaves a silvery appearance. Flowers are borne in clusters of one to three, originating from leaf axils in the upper portion of stems. Flowers are white, sometimes with a hint of yellow, have purple marks and bloom from July through October. Chinese lespedeza is capable of both cross-pollination from other plants and self-pollination. After flowers are pollinated, small flat to rounded pods containing a single seed are produced in terminal axils. As the pods mature they turn from green to brown and eventually drop their seeds, which may remain viable for 20 years.

(top) Flowering lespedeza showing trifoliate leaves. (middle) Lespedeza plants. (bottom) A meadow infested by lespedeza, flowering in September. Photos by Adam Gundlach, Wildlife Habitat Council

Chinese lespedeza was introduced to the United States by various state and federal agencies for erosion control, soil improvement, and as potential wildlife forage. It primarily invades open habitats such as meadows, prairies, open woodlands, and fields. It is a hardy species that tolerates conditions many native species do not, including infertile soils and drought conditions. Once established, it forms an extensive perennial root system and seed bank that makes it difficult to eradicate. New growth begins each spring from root crown buds near the base of last year's stem. Dense stands of Chinese lespedeza crowd out native plants and develop abundant soil seed banks that ensure its continued presence in an area if no control measures are taken.

Control – Hand removal is not effective due to the deep perennial root system produce by Chinese lespedeza. Repeated mowing for many years may suppress its spread, but infestations can return if mowing is discontinued prior to root reserves being exhausted. The most effective control method will likely be a combination of early season mowing followed by an herbicide treatment to the new sprouts. Both mowing and herbicide applications should be performed from early to mid summer, prior to the flower bud stage. Herbicides known to be effective against *L. cuneata* include metsulfuron methyl, triclopyr, clopyralid, and glyphosate. Prescribed burns may also be incorporated into control regimes, but burning alone stimulates resprouting and seed germination. To be effective, burns must be followed by herbicide application to resprouts.

Lespedeza stems in April with seeds still attached from previous year. Photo by Adam Gundlach, Wildlife Habitat Council.

NOTE: *Lespedeza cuneata* may be confused with native *Lespedeza* species, including slender bushclover (*Lespedeza virginica*). *L. cuneata* is the only member of the genus that has wedge-shaped leaf bases. It has abundant, branching, coarse stems that distinguish it from the fewer weak stems produced by *L. virginica*. It is extremely important to properly identify the species prior to initiating management efforts.

Compiled from the PCA Alien Plant Working Group Fact Sheet, Plant Invaders of Mid-Atlantic Natural Areas, and Miller 2003.

Purple loosestrife (*Lythrum salicaria*)

Description & Biology – Purple loosestrife is a highly invasive perennial herb native to Eurasia. It invades a variety of wetland habitats. Purple loosestrife's erect, square, semi-woody stems grow between four and ten feet tall. Plants are generally covered with fuzzy hairs, and mature plants can produce up to 50 stems from a single root crown. The lance-shaped leaves occur in an opposite or whorled arrangement, do not possess stalks, and are rounded or heart-shaped at the base. Purple loosestrife blooms from June through September, producing a showy display of purple to magenta flowers that are borne on terminal spikes at the ends of stems. Mature plants can have thousands of flowers that give rise to seed capsules, with each capsule containing more than 100 seeds on average. A single plant may produce between two and three million seeds in a single growing season. Purple loosestrife reproduces mainly by seed but can also spread vegetatively from its hardy, lateral-branching rootstock and plant fragments, which can root to form new plants.

Purple loosestrife flower stalk. Photo by Adam Gundlach, Wildlife Habitat Council.

Purple loosestrife invades a variety of wetlands habitats including marshes, river and stream banks, wet meadows, pond margins, and ditches. It achieves peak growth in full sun and moist soil but can tolerate locations with up to 50 percent shade. Purple loosestrife infestations alter hydrological processes, nutrient cycles, and biogeochemistry of wetlands. Its high reproductive output allows it to be highly competitive in wetland habitats, displacing native vegetation that is of greater value to wildlife. Many obligate marsh birds avoid nesting in marshes that have been invaded by purple loosestrife.

(above left) Purple loosestrife foliage in late June. Photo by Adam Gundlach, Wildlife Habitat Council. (above right) Purple loosestrife infestation. Photo by Randy Westbrooks, U.S. Geological Survey, Bugwood.org.

Control – Small patches can be hand pulled or dug, but all plant fragments must be removed from the site to prevent sprouting. The extensive seed bank will continue to regenerate for many years, making continued monitoring and control a necessity. Purple loosestrife should not be cut or mowed, as these techniques only serve to spread plant fragments that can sprout vegetatively and increase an infestation. Herbicide application, typically wetland-approved glyphosate, is effective for large infestations, either as a cut-stump method for small patches or a foliar spray for large infestations. For the cut-stump method, stems should be cut about six inches above the ground and herbicide should be applied directly to the cut surfaces. All cut vegetation should be removed from the site. Herbicide applications are most effective when the plants are approaching dormancy; however, a mid-summer application followed by a late-season application will likely reduce the amount of seed produced. It is critical to use herbicide that is registered for use in wetland habitats, such as Rodeo®.

Flowering purple loosestrife plants. Photo by John D. Byrd, Mississippi State University, Bugwood.org.

Biological control is seen as the most appropriate method for long-term control of large infestations. Four biocontrol agents have been released throughout the eastern United States, including three weevil species (*Galerucella calmariensis*, *Galerucella pusilla*, *Hylobius transversovittatus*) and one beetle species (*Nanophyes marmoratus*). All four of the introduced species have become established and have exhibited effective suppression of purple loosestrife populations, with the early release sites showing the best results.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and Southeast Exotic Pest Plant Council Plant Manual.

Japanese stiltgrass (*Microstegium vimineum*)

Description & Biology – Japanese stiltgrass, also called Nepalese browntop, is a sprawling annual grass native to southern and southeastern Asia. It has wiry, alternately-branched stems that may reach four feet in length. Leaves are green, lance-shaped, with few hairs on the upper and lower surfaces, and about four inches long. The leaves have a distinctive midrib and are asymmetrical with respect to the midrib. A small, inconspicuous flower stalk is produced in late summer, and flowers bloom through September. Seeds are produced in a terminal sheath-like structure soon after flowering and the entire plant dies by the fall. A single plant may produce up to 1,000 seeds that can remain viable in the soil for three years or more. It reproduces solely by seed, which is dispersed near the parent plant and transported greater distances by water, animals, and human activity.

(top) Young stiltgrass leaves in May. (above) Stiltgrass infestation in forest understory in September. Photos by Adam Gundlach, Wildlife Habitat Council.

Japanese stiltgrass is a shade-tolerant grass that typically inhabits flood plains, stream banks, mesic woodlands and fields, recreational trails, roadsides, lawns, and other areas of disturbance. Due to its shade tolerance – growing in as little as five-percent full sunlight - stiltgrass readily dominates forest understories, displacing native plant communities and altering soil chemistry and organic matter composition. It is a colonial species and individual plants can spread by rooting at stem nodes that contact soil. It can quickly take over disturbed areas, forming dense monotypic stands, and can slowly invade undisturbed areas over several years. Abundant deer populations can exacerbate the problem by selectively foraging native vegetation while avoiding the unpalatable stiltgrass. Stiltgrass can often be found growing along recreational trails, which facilitate dispersal of seeds to new locations.

Control – Hand pulling is an option for small infestations. Pulling in mid- to late-summer when the plants are larger makes it easier to grasp clumps of stiltgrass at the base of the stem. Removal should occur from late summer through early November before seed set to prevent dispersal of seeds. If removal occurs after fruit has been produced, all plant material should be bagged and properly disposed offsite. Hand removal performed too early in the season (before July) may allow seeds in the soil to germinate, mature, and produce seeds by

(above left) Close up of stiltgrass leaves in late July. (above right) Stiltgrass leaves and apical flower stalk (arrow) protruding from leaf sheath, late September. Photos by Adam Gundlach, Wildlife Habitat Council.

the end of the growing season. It is important to avoid damaging native grasses and forbs while pulling. Many other plants may be interspersed with the stiltgrass. For large infestations, herbicide may be the only viable control option. Glyphosate is effective, but it is non-specific and will kill desirable vegetation. Grass-specific herbicides such as fluazifop-p (Fusilade®), imazameth (Plateau®), and sethoxydim (Poast®, Vantage®) control stiltgrass and allow other native vegetation to remain onsite. Control measures need to be repeated for several years (up to seven) until the seed bank is exhausted.

Compiled from the PCA Alien Plant Working Group Fact Sheet, Southeast Exotic Pest Plant Council Plant Manual, The Nature Conservancy Element Stewardship Abstract, and Van Driesche et al. 2002.

Chinese silvergrass (*Miscanthus sinensis*)

Description & Biology – Chinese silvergrass is an ornamental grass native to Southeast Asia that was introduced to the United States around 1900 for landscaping purposes. It is still used today as an ornamental species for landscaping and along roadsides as a visual barrier. Silvergrass is a hardy perennial grass that forms dense clumps from five to ten feet tall. The long, slender basal leaves reach about three feet or more in length and have a white midrib. There are several cultivars of *Miscanthus* that range in leaf color from green to a combination of green and white. Flowers are produced from late summer into the fall and are borne as a showy terminal panicle (multi-branched flowering stalk). The panicle can reach two feet in length and is silvery in coloration, sometimes with a hint of pink. The flower stalk persists through the winter, and seeds develop from September through January. Although it is believed that seeds of most silvergrass varieties are viable, once established in an area, it mainly reproduces vegetatively through new shoots that arise from rhizomes.

Chinese silvergrass with flower stalks. Photo by James H. Miller, USDA Forest Service, Bugwood.orgl

Chinese silvergrass grows best in sunny areas with rich, moist, well-drained soils. It has escaped ornamental cultivation and tends to invade roadsides, forest margins and openings, shorelines, abandoned fields, and other disturbed areas. *Miscanthus* poses a fire hazard due to its large, feathery flower plumes and large amount of highly flammable above ground biomass. Burning silvergrass can be difficult to control because it produces tall flames and sends up burning plant fragments that can jump fire lines.

Chinese silvergrass with fluffy seed-bearing plumes. Photo by Chris Evans, River to River CWMA, Bugwood.org

Control – The hardy root system produced by silvergrass makes it difficult to control, as the entire root system must be killed to prevent resprouting. New plants may develop from rhizome fragments. Young plants can be dug or hand-pulled, but care needs to be taken to remove all root fragments. Mature plants are most effectively controlled by herbicide application – glyphosate (e.g. Accord, Roundup Pro) or imazapyr (e.g. Arsenal) – applied in late spring to early summer or the fall. The plant must be actively growing for herbicide to be effective. Control areas need to be monitored regularly to remove any resprouts that develop in subsequent years.

Compiled from the Southeast Exotic Pest Plant Council Plant Manual, Plant Invaders of Mid-Atlantic Natural Areas, and Miller 2003.

Wavyleaf basketgrass (*Oplismenus hirtellus* ssp. *undulatifolius*)

Description & Biology – Wavyleaf basketgrass is a perfect, if unsettling, example of why vigilant monitoring is the foundation of invasive species management. This southeast Asian grass, whose leaves have a rippling pattern reminiscent of waves coming into shore, is on the front line of invasive species control in the Chesapeake Bay Watershed. There are no records of its presence in the United States until 1996, when botanists noticed the unusual-looking grass growing in a Maryland state park near Baltimore. Since then, scientists discovered at least half a dozen more populations, including two in Virginia. Government agencies and nonprofit/research organizations are running an intensive Early Detection and Rapid Response effort in hopes of eradicating the grass from the continent while it is still contained in a small geographic area. This effort includes training volunteers and conservation professionals to recognize and remove the grass, as well as mapping and monitoring its distribution. The Maryland Department of Natural Resources invasive plant specialists are asking anyone who sees this grass to notify them (DNR Wildlife and Heritage Service, 301-948-8243).

Identifying chracteristics of wavyleaf basketgrass include: hairy stems, leaves that have a rippling pattern reminiscent of waves coming into shore, and a tendency to trail across the ground rather than grow upright. Photo by Kerrie Kyde, Maryland DNR.

Wavyleaf basketgrass creeps across the ground, rooting at nodes on its hairy stems. Its tendency to lay nearly flat on the ground is one characteristic that helps distinguish it from look-alike species. This perennial grass invades forested areas, especially moist areas along waterways, though it appears to also tolerate dry conditions. Leaf blades are ½ to one inch wide and 1½ to four inches long. Seedheads appear in fall (generally mid-September through November in the Chesapeake Bay Watershed) and are characterized by bunches of spikelets that have glumes and long, sticky, bristle-like appendages called awns.

In the southernmost portions of the Chesapeake Bay Watershed, a native subspecies of basketgrass, *Oplismenus hirtellus* ssp. *setarius*, exists. The native and non-native subspecies can be distinguished by the presence or absence of hairs on the stems: the native has hairless or nearly hairless stems, while the non-native invasive has many hairs, as shown in the picture above. Another grass that can be confused with wavyleaf basketgrass is small carpgrass (*Arthraxon hispidus*), an exotic species introduced from the eastern hemisphere. Small carpgrass has wavy leaves similar to those of wavyleaf basketgrass, but small carpgrass grows upright rather than creeping horizontally along the ground, and its seedheads are drooping clusters of about five branches (somewhat resembling small hands), whose spikelets lack the bristly awns that characterize wavyleaf basketgrass seedheads.

Wavyleaf basketgrass seedheads in fall. Sticky awns allow seeds to move via animals and shoes. Photo by Kerrie Kyde, Maryland DNR.

Small carpgrass is prohibited as a potentially invasive species in some northeastern states, but it does not appear to threaten native ecosystems anywhere near as severely as wavyleaf basketgrass does. Other species that may be confused with wavyleaf basketgrass are the native deertongue (*Dichanthelium clandestinum*), which has similar hairy stems but grows upright and does not have wavy leaves, and the non-native invasive Japanese stiltgrass (*Microstegium vimineum*) which invades similar habitats but grows upright and has smooth leaves with a silver stripe down the middle. Page 23 contains more information about Japanese stiltgrass.

Control – Anyone who sees this grass is asked to immediately contact the Maryland DNR (Wildlife and Heritage Service, 301-948-8243). The experts at the agency and its partner organizations can help determine appropriate control actions, which may include hand pulling and foliar spray. During the fall, signs should be erected urging anyone passing through an invaded area to thoroughly remove seeds and other plant matter from shoes, socks, equipment (e.g. bike tires), and pets before entering another natural area. The DNR experts can advise whether the signs should tell passersby to brush the seeds onto the ground in the invaded area or bag them and put them in the trash. With EDRR efforts, it is critical to keep the species contained geographically, so while it may seem counterintuitive, experts sometimes recommend brushing seeds onto the ground rather them putting them in the trash and allowing for the possibility of starting a new population in a landfill.

Compiled from the Maryland DNR information page, Kerrie Kyde (habitat ecologist/ invasive plant specialist, Wildlife and Heritage Service, Maryland DNR, personal communication 21 January 2009), Marc Imlay (conservation biologist, Anacostia Watershed Society, personal communication throughout 2008 and January 2009), and Dr. Paul Peterson (research scientist & curator of grasses, Smithsonian Museum of Natural History, personal communication July 2008).

Japanese spurge (*Pachysandra terminalis*)

Description & Biology – Japanese spurge is a perennial evergreen herb native to Japan that is used as a ground cover in landscaping. Stems grow to about one foot in height with alternate leaves that are generally grouped near the apex of stems. The leaves are dark green (except for variegated forms), shiny, oval to teardrop-shaped, and are coarsely serrated toward the tip. Small white tubular flowers are borne in a terminal spikes and bloom in early spring from March through April. The small light green fruit produced is not an important source of reproduction; the main form of reproduction is through spreading rhizomes that readily send up new shoots. Rhizome fragments can root to form new plants.

Japanese spurge grows in locations with partial to full shade and grows best in areas with rich, moist, well-drained soil. Once established, it can easily spread by rhizomes to form a dense ground cover that excludes other plant species. It typically invades forests and meadow margins but will not grow in areas that receive full sun.

Control – Small patches can be removed by hand. Care must be taken to remove as much of the root system as possible. Herbicides such as glyphosate are effective for larger infestations. Monitoring and additional control are necessary in subsequent years, especially after hand pulling, because resprouts emerge from rhizome fragments.

(top) Japanese spurge foliage and flower stalk. (above) Typical spreading growth of Japanese spurge. Photos by Jil M. Swearingen, USDI National Park Service, Bugwood.org

Compiled from the U.S. Forest Service Weed of the Week.

Reed canary grass (*Phalaris arundinacea*)

Description & Biology – Reed canary grass is a perennial, cool-season, sod-forming grass that is native to Europe and potentially Asia and temperate portions of North America – though the latter two are still debated. It produces erect, hairless stems (culms) that grow from two to nine feet in height. Long, slender leaves grow from three inches to more than a foot long and are rough on both the upper and lower surfaces. Flowers occur in dense clusters (panicles) at the apex of the stem. Young flowers start off green or purple in color and transition to tan or beige as the flowers mature. The shiny brown seeds ripen in late June and are dispersed by water, wildlife, and human activity. Creeping rhizomes capable of rooting at the nodes send up numerous shoots that facilitate the prolific spread of reed canary grass.

(top) Flowering reed canary grass. Photo by Chris Evans, River to River CWMA, Bugwood.org. (middle) Stem and leaves. Photo by Richard Old, XID Services, Inc., Bugwood.org. (above) Reed canary grass infestation. Photo by Michael Shephard, USDA Forest Service, Bugwood.org

Reed canary grass generally grows on moist, fertile soils, often invading riparian corridors and other wetland habitats such as wet meadows, marshes, low-lying fields, floodplains, and ditches. It also is capable of growing on dryer upland sites, including partially shaded areas, and tolerates periods of inundation and freezing temperatures. It readily invades disturbed areas and over time can form persistent monotypic stands that displace and prevent regeneration of other vegetation.

Control – Dense stands are difficult to eradicate because of persistent rhizomes and an abundant soil seed bank. A combination of treatments often produces the best control results. The most effective control method for large infestations is herbicide application. Applicators must be sure to only use herbicides that are labeled for use in aquatic environments, such as Rodeo®, as reed canary grass often inhabits wetland areas. Following herbicide application, the dead leaf litter can be mowed to encourage other plants to germinate and grow.

Repeated mowing three to five times per growing season may reduce the vigor of reed canary grass rhizomes but often is not effective in eradicating an infestation. Covering the infested area with dark plastic sheeting (called soil solarization) for several months may kill the grass, though results have been mixed. The plastic should be tightly secured around the edges and no shoots should be allowed to emerge from the edges of the plastic, because exposed shoots allow energy to be transferred to the rhizome system.

Soil erosion following removal of reed canary grass may be a concern. In addition, reed canary grass readily invades bare soil, so management areas should be promptly planted with native vegetation to reduce the likelihood of erosion or re-infestation occurring.

Compiled from the U.S. Forest Service Weed of the Week, and The Nature Conservancy Element Stewardship Abstract.

Common reed (*Phragmites australis*)

Description & Biology – Common reed is a tall, perennial, clonal grass with various sub-species found around the world. Though a native sub-species of common reed (*Phragmites australis* ssp. *americanus*) has existed in North America for thousands of years, non-native sub-species were introduced by early colonists and have recently experienced rapid expansion throughout the Northeast and Midwest United States and along the Atlantic coast. The introduced *Phragmites* forms dense stands with hollow, woody stems (culms) that grow to more than 15 feet in height. Leaves are slender and pointed, reaching from 8 to 16 inches long with sheaths that wrap around the stem. Flower clusters develop in mid-summer as terminal panicles. Flowers are generally golden to purplish in coloration, and as seeds mature they take on a fluffy grey appearance from hairs on the seeds. An individual plant can produce thousands of seeds each year, though a large quantity is not viable. *Phragmites* reproduces mainly vegetatively through extensive rhizomes. Roots and rhizomes may extend several feet below the ground, dominating both space and soil nutrients. Buds produced on the rhizomes send up shoots that develop into new above ground growth. Rhizome fragments can sprout to form new plants and can be transported by water or other means to colonize new locations.

(top) Flowering *Phragmites* in July. (above) *Phragmites* stem and leaves. Photo by Adam Gundlach, Wildlife Habitat Council.

Phragmites inhabits a variety of wetland habitats including freshwater marshes, riparian areas, tidal and nontidal brackish environments, lakeshores, roadside ditches, and most any mesic to wet site. It can quickly invade areas of disturbance but will also invade undisturbed sites and can tolerate alkaline, saline, and acidic conditions. The introduced varieties form dense monocultures that displace native vegetation and alter ecosystem functions by dominating water, space, and nutrients. This process results in degraded wetland habitats with diminished biodiversity. After being established for several years, a dense leaf litter develops under *Phragmites* stands, further restricting growth of other vegetation.

Control – Implementing control efforts early in an infestation allows for easier control. Small patches of *Phragmites* that do not exhibit expansion into surrounding areas should not be a major management concern. For patches with invasive tendencies, mowing repeatedly for several years may stop the spread but is not likely to kill the plant. Large infestations are best controlled with herbicide treatment. Glyphosate products registered for use in wetland habitats (Rodeo®) are the most effective at controlling dense, established stands. Herbicides are most effective when applied in late summer or early fall after flower tassels have developed. The plants allocate nutrients to the root system during this period. Prescribed burns performed late in the growing season after the flower tasseling stage can be somewhat effective when used in combination with herbicide treatments. Burning removes the leaf litter and promotes germination of native plants. Prescribed burns used alone are not effective at controlling *Phragmites*.

Phragmites plants in July. Photo by Adam Gundlach, Wildlife Habitat Council.

(above left) *Phragmites* stand in a sandy beach along the Chesapeake Bay, early August. (above right) Dormant *Phragmites* towering above the surrounding cattails in January. Photos by Adam Gundlach, Wildlife Habitat Council.

Compiled from the PCA Alien Plant Working Group Fact Sheet, The Nature Conservancy Element Stewardship Abstract, and Van Driesche et al. 2002.

Golden bamboo (*Phyllostachys aurea*)

Golden bamboo infestation. Photo by Adam Gundlach, Wildlife Habitat Council

Description & Biology – Golden bamboo is native to China and has been cultivated in surrounding countries for centuries. It was introduced to the United States in the late 1800s and continues to be commercially available today. Its jointed canes, or main stems, can grow to heights of more than 30 feet, with leaf stalks borne singly at each node on the cane. The grass-like leaves are lanceolate, about six inches long, and can be rough or smooth along the edge. Golden bamboo rarely flowers – it may not flower for several decades – but when it does, it produces flower spikelets containing 8 to 12 flowers. Its main form of reproduction is through rhizomes that spread from the parent plant and produce abundant new above ground shoots. Generally, after golden bamboo produces flowers, a portion of the plant dies back, leaving standing dead canes.

Golden bamboo grows best in locations with full sun and moist loamy soil, but it can slowly invade forests with sparse canopy or other less-than-ideal locations. Once established, its vigorous rhizomatous growth can quickly form dense, monotypic stands that prevent growth of other vegetation.

(above left) Young bamboo (on left side) next to mature culms, July. (above right) Close up of mature culms, early August. Photos by Adam Gundlach, Wildlife Habitat Council

Control – Cutting or mowing golden bamboo multiple times per growing season for several years will reduce energy stored in the rhizomes and result in the eventual death of the plant. For more immediate results, large infestations can be treated with a foliar herbicide spray. In locations where golden bamboo is growing among other desirable plant species, a cut-stump application of systemic herbicide (glyphosate) will reduce negative impacts on surrounding vegetation.

Golden bamboo leaves with culms in background, early August.
Photo by Adam Gundlach, Wildlife Habitat Council

Compiled from the U.S. Forest Service [Weed of the Week], and Southeast Exotic Pest Plant Council [Plant Manual].

Japanese knotweed (*Polygonum cuspidatum*)

Description & Biology – Japanese knotweed, also known as crimson beauty, Mexican bamboo, or Japanese fleece flower, is a shrub-like, herbaceous, perennial plant native to eastern portions of Asia. Its dense, thicket-forming growth can reach ten feet tall. The leaves vary in size and shape but are generally broadly oval, about six inches long and three inches wide, and pointed at the tip. Stems are smooth, erect, and swollen at leaf axils, and they often have a zig-zag appearance. Small white to greenish-white flowers are borne on densely clustered spikes that arise from the leaf axils and bloom from August through September. Japanese knotweed is dioecious, with male and female flowers occurring on separate plants. Male flowers typically point upward, while female flowers droop down. Female plants produce winged fruits that contain small, shiny, triangular seeds (called achenes). The seeds do not appear to be an important means of reproduction in the United States. Japanese knotweed grows stout rhizomes that can extend 15 to 20 feet and readily produce new shoots. Japanese knotweed is a hardy species that tolerates harsh conditions such as salinity, drought, and high temperatures, as well as a variety of soils. It generally grows in locations that receive full sun and has not yet been observed to invade forest understory. Once established, it quickly forms dense thickets that displace native vegetation and alter ecosystem functions. Its early emergence in the spring and rapid growth rate give it a competitive advantage over native plants. It often invades shorelines along streams and rivers, waste places, rights-of-way, and other areas of disturbance. It is able to persist in areas prone to severe flooding because its seeds and rhizomes are dispersed by floodwater and can rapidly colonize an area scoured by flooding.

Japanese knotweed foliage and stem, mid August. Photo by Adam Gundlach, Wildlife Habitat Council.

Control – Japanese knotweed can persist at a site for long periods because of its extensive rhizomes. Young plants may be hand pulled given immature roots and loose soil conditions. Small patches may be grubbed using a weed wrench or similar tool to remove the entire plant including all roots and rhizomes. Any root fragments remaining in the soil can sprout to form new plants. All vegetative matter should be bagged and properly disposed offsite to prevent resprouting. Various herbicides can be employed to control Japanese knotweed and applied either as a cut-stem treatment or as a foliar application. For sensitive areas and situations where non-target species are intermixed with Japanese knotweed, the cut-stem

Typical zig-zag growth of Japanese knotweed stems. Photo by Jack Ranney, University of Tennessee, Bugwood.org.

application method can be used in conjunction with glyphosate (e.g. Accord) or triclopyr (e.g. Garlon). Since this species often occurs in riparian areas, it is crticial to use a wetland-approved chemical whenever runoff into water is possible. sFor extensive, monotypic stands of knotweed or locations where non-target damage is acceptable, a foliar application of glyphosate or triclopyr is most effective. No matter which method is employed, treatments need to be repeated for multiple seasons to eliminate all seedlings and rhizome sprouts.

Japanese knotweed flowering. Photo by Leslie J. Mehrhoff, University of Connecticut, Bugwood.org.

Compiled from the PCA Alien Plant Working Group Fact Sheet, The Nature Conservancy Element Stewardship Abstract, and Southeast Exotic Pest Plant Council Plant Manual.

Lesser celadine (*Ranunculus ficaria*)

Description & Biology – Also known as fig buttercup, many varieties of this European native were introduced to the United States for ornamental purposes. It is a low-growing, perennial plant with shiny, dark green leaves that are kidney-shaped to heart-shaped. The leaves develop during the winter and generally die back by June. Glossy, bright yellow flowers are borne on slender stalks extending above the basal leaves and bloom from late winter through May. Late in the flowering period, small pale bulblets are produced on leaf stalks. The plant also develops abundant underground tubers, capable of forming new plants when separated from the parent plant. Both the bulblets and tubers can be scattered locally by wildlife or carried to new locations by water.

Lesser celadine flowers and foliage. Photo by Leslie J. Mehrhoff, University of Connecticut, Bugwood.org

Fig buttercup prefers moist forest understory conditions and flood plains. It forms thick carpets that blanket the forest floor and prevent growth of other vegetation. It is especially detrimental to native spring wildflowers and their related organisms. Fig buttercup's early growth period allows it to become well established before native spring plants begin growth.

NOTE: Fig buttercup is similar in appearance to native marsh marigold (*Caltha pulstris*), which has long flower stalks (eight inches or more), does not form a dense mat of growth, and does not produce bulblets or tubers. It is important to carefully identify the species prior to initiating control measures.

Control –Small patches of fig buttercup can be hand pulled or dug, but all bulblets and tubers must be removed to prevent new sprouts, which can be difficult. Large infestations are most effectively controlled with systemic herbicide, such as glyphosate, which can be applied in late winter or early spring as long as temperatures remain above 50°F. The herbicide application should be performed as early as possible to avoid damage to native spring species. It is important to avoid application to non-target vegetation, as glyphosate is a non-specific herbicide. Amphibians that emerge in early spring should also be considered when planning an herbicide application, as they are highly sensitive to such chemicals; applications should be made prior to their emergence.

Lesser celadine infestation. Photo by Leslie J. Mehrhoff, University of Connecticut, Bugwood.org

Compiled from the PCA Alien Plant Working Group Fact Sheet, and Plant Invaders of Mid-Atlantic Natural Areas.

Japanese bristlegrass (*Setaria faberi*)

Description & Biology – Also known as giant foxtail, Chinese millet, and nodding foxtail, among other names, Japanese bristlegrass is a summer annual grass native to Asia that can be found throughout much of the United States. Its stems grow from two to five feet tall, often branching near the base of the plant so individual plants have multiple stems. The floppy leaves grow from five to 20 inches long and about one-half-inch wide. The leaves are covered in fine hairs and the leaf margin is rough due to tiny serrations. Each stem ends in a terminal panicle of clustered florets, with each floret possessing a single stiff hair that

(above left) Japanese bristlegrass. Photo by Leslie J. Mehrhoff, University of Connecticut, Bugwood.org. (above right) Japanese bristlegrass flower head. Photo by Dan Tenaglia, Missouriplants.com, Bugwood.org

forms the distinctive bristles of the plant. The wind-pollinated flowers bloom from late summer into early fall. Flower heads can reach seven inches long and often begin to droop at this size, hence the common name nodding foxtail. Japanese bristlegrass reproduces strictly by prolific seed production.

Japanese bristlegrass invades cropland, fields, meadows, roadsides, waste areas, pastures, and forest edges. It is often found on sunny mesic to dry sites, growing best in fertile sandy locations, but also adapts to a variety of environmental conditions.

Control – Japanese bristelgrass can be removed by hand, dug up, or tilled in late summer prior to seed set. Because the seeds remain viable for approximately two years, preventing seed production for several consecutive years should effectively eradicate the plant from a site. A variety of herbicides are also effective at controlling Japanese bristlegrass, but the appropriate herbicide varies depending on site-specific conditions. The use of fire as a control method is not recommended, as bristlegrass infestations have been noted to increase following fire disturbance.

Bristlegrass in December. Photo by Adam Gundlach, Wildlife Habitat Council

Compiled from the U.S. Forest Service [Weed of the Week](#).

Johnsongrass (*Sorghum halepense*)

Description & Biology – Johnsongrass is a perennial grass native to the Mediterranean region that forms dense clumps or solid stands of growth reaching eight feet or more in height. The smooth, lanceolate leaves reach two feet long and possess a distinct white mid-vein stripe. The stout stems are smooth and are sometimes pink or rusty near the base. Flowers are borne on open, whorled branching, terminal panicles that generally appear slightly purplish or brown. Flowers begin to bloom approximately two months following initiation of growth in the spring. Flowers can be self pollinated in the absence of conspecific plants, ensuring the production of seed. Abundant brown seeds mature from May through March. Reproduction is accomplished through seed and rhizomes. Johnsongrass produces an extensive network of rhizomes that readily sprout when fragmented to form new plants. The rhizomes and ample seed bank allow johnsongrass to persist at a site for many years. Seeds may remain viable for five years or more and can be dispersed long distances by water, animal movement, and human activity.

(above left) Johnsongrass florets. Photo by James H. Miller, USDA Forest Service, Bugwood.org. (above right) Flowering plant. Photo by Charles T. Bryson, USDA Agricultural Research Service, Bugwood.org

Johnsongrass tends to occur in old fields, rights-of-way, forest openings and edges, ditches, pastures, and along wetlands. It grows best in disturbed open sites with rich soil, especially agricultural fields and their margins. The aggressive colonial spread of johnsongrass and its ability to survive in a range of environmental conditions make it one of the worst weed species in the world, occurring in 53 countries. It significantly reduces tree seedling survival in natural forest regeneration and tree plantations.

Control – Small isolated occurrences of johnsongrass can be pulled or dug up when soil is moist and soft following rain. All vegetative material should be bagged and properly disposed, as any remaining stem or rhizome fragments can sprout to form new plants. Larger areas can be repeatedly mowed close to the ground for several growing seasons to deplete energy reserves in the rhizomes and prevent seed development. Tilling can also be used for control, but it may increase an infestation by breaking up rhizomes and stimulating new sprouts to develop if not repeated throughout the growing season.


Johnsongrass infestation. Photo by Adam Gundlach, Wildlife Habitat Council

Non-specific systemic herbicides, such as glyphosate, effectively kill rhizomes and above ground growth. In locations where damage to native seedlings and other vegetation is a concern, a grass-specific herbicide could be employed. Herbicide applications will likely be required for multiple years to ensure that the seed bank and rhizomes have been exhausted.

(above left) Pointed rhizomes extending laterally from johnsongrass roots. (above right) Stem and leaf with distinct white mid-rib and view of the ligule at its base. Photos by Steve Dewey, Utah State University, Bugwood.org.

Compiled from the U.S. Forest Service Weed of the Week, The Nature Conservancy Element Stewardship Abstract, and Invasive Plants of the Eastern U.S.

INVASIVE VINES

Adapted from PCA Alien Plant Working Group – Mid-Atlantic List

Five-leaf akebia (*Akebia quinata*)

Description & Biology – Also called chocolate vine, this ornamental vine was introduced to the United States from East Asia in the 1800s. It is a perennial, deciduous to evergreen (in warmer climates) vine found throughout much of the eastern United States. The trailing or climbing vines are slim, round, and green when young, becoming brown as the vine matures. Its palmately compound leaves alternate along the stems and are each composed of five oval leaflets. Five-leaf akebia blooms in late March to early April and produces fragrant, chocolate-purple flowers, although other cultivated varieties produce white flowers. The fruit is a flattened, violet pod, two to four inches long, that ripens from September to October, splitting in half to reveal a white, cocoon-like core containing numerous black seeds. Five-leaf akebia spreads locally through an extensive system of rhizomes that send out new shoots. Its seeds can be dispersed longer distances to colonize new locations.

Photo by Shep Zedaker, Virginia Polytechnic Institute and State University, Bugwood.org

Five-leaf akebia is both shade and drought tolerant, which gives it the ability to invade forest environments. After becoming established, it can quickly form a dense carpet of vines in the understory that, if left unchecked, can eventually twine their way into the canopy and overtop trees and shrubs. The dense growth produced by established infestations prevents seed germination and seedling development of native plant species. Under optimal conditions, vines may grow up to 40 feet in a growing season.

Control – Five-leaf akebia can be controlled through a variety of methods, depending on the site conditions and available resources. A combination of methods will likely be most effective. For small patches, cutting or pulling combined with removal of the roots can be effective if performed for several years. Climbing vines should be cut at ground level. Extensive infestations can be treated with systemic herbicides, such as glyohosate or triclopyr, in areas where allowable. Herbicides can be applied as foliar, cut-stem, or basal bark applications. Foliar applications are most efficient for treating extensive infestations but are likely to impact non-target species as well. The cut-stem method is the most selective and thus best for avoiding damage to non-target species, but this method is time intensive and not practical for large areas.

Five-leaf akebia infestation. Photo by Shep Zedaker, Virginia Polytechnic Institute and State University, Bugwood.org

Compiled from the PCA Alien Plant Working Group [Fact Sheet], U.S. Forest Service [Weed of the Week], and [Plant Invaders of Mid-Atlantic Natural Areas].

Porcelainberry (*Ampelopsis brevipedunculata*)

(top) Porcelainberry leaves, stem, and flowers and young berries forming. (above) Highly variable leaf forms exhibited by porcelainberry. Photos by Adam Gundlach, Wildlife Habitat Council

Description & Biology – Porcelainberry is a highly invasive, perennial vine originating from northeastern Asia. It closely resembles native grape species in the genus *Vitis* and can also be confused with native species in the genus *Ampelopsis*. Its deciduous leaves take on a variety of shapes from a typical maple leaf form to a deeply lobed form with lobes extending in to the leaf veins. The leaves are alternately arranged on woody stems. Non-adhesive tendrils develop opposite of leaves and allow porcelainberry to twine around and climb over other vegetation. Small greenish-white flowers also occur opposite the leaves, blooming from June through August. Colorful berries develop in September and October. As the berries mature they change colors from purple to green, to bright blues. The berries are eaten by birds and small mammals and then dispersed to new locations. Seeds remain viable in the soil for several years. Porcelainberry also reproduces vegetatively in locations where it is already established, as its large taproot sends up new vines following cutting or other trauma.

Porcelainberry grows well in many soil types and typically is found in forest edges and openings, pond margins, riparian areas, flood plains, and other disturbed areas. Locations with full shade or permanent inundation discourage growth of porcelainberry. New stands are often found downstream from established infestations, suggesting that seeds are transported by water.

(above left) Porcelainberry infestation in early June. (above right) Foliage and maturing berries, late September. Photos by Adam Gundlach, Wildlife Habitat Council.

Control – Small patches with young plants can be hand pulled or dug. Any plants that have already produced fruit should be bagged and properly disposed. Larger vines can be cut near their base. Cutting promotes sprouting from the root system and needs to be repeated several times during the growing season. Cut-stump applications of herbicide discourage resprouts and increase the effectiveness of treatments. Large infestations can be treated with systemic herbicide, such as triclopyr or glyphosate, either as a foliar application or as a basal bark application. Control measures need to be repeated for several years to ensure all plants have been killed and the seed bank has been exhausted.

NOTE: Some porcelainberry leaf forms resemble native grape vine (*Vitis* spp.), and the two species may be found growing intermixed. Two native species of *Ampelopsis* (*A. arborea* & *A. cordata*) also occur in the eastern and central United States. It is important to ensure proper identification before initiating management. The photo above depicts a porcelainberry leaf in the lower right next to grape leaves in the upper left. Photo by Adam Gundlach, Wildlife Habitat Council.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and U.S. Forest Service Weed of the Week.

Oriental bittersweet (*Celastrus orbiculatus*)

Description & Biology – Native to Eastern Asia, oriental bittersweet is a deciduous, woody, perennial vine. The alternate leaves are nearly round, variable in size, and sometimes come to a point at the tip, with finely toothed margins. The stems are round, light brown, and smooth with small light spots, which are lenticels that aid in moisture and gas exchange. Oriental bittersweet is dioecious, meaning female and male flowers are produced on separate plants. Female plants produce numerous clusters of small green flowers that are borne in most leaf axils. Flowers bloom in May and are pollinated by insects and wind. Fruits are green during early development and become a bright yellowish orange as they mature. The fruits ripen in August and September and split open to reveal fleshy, red-orange, seed-containing pods that remain on the plant through the winter. In addition to seed production, established infestations expand through root suckering.

Oriental bittersweet grows in a variety of habitats including forest and forest edges, fields, thickets, roadsides, hedgerows, and other areas of disturbance. It is shade tolerant, allowing it to germinate and grow under complete canopy cover. Its vigorous growth often overtops and smothers other vegetation or shades out plants growing below. As it climbs into the canopy, the increased weight may lead to uprooting of trees during heavy winds or snow. The seeds of oriental bittersweet readily germinate and remain viable for several years. Once established, the prolific seed production quickly creates a persistent seed bank. Oriental bittersweet is displacing American bittersweet (*Celastrus scandens*) in many areas through direct competition and hybridization.

Control – Vines can be cut near the root collar to kill above ground growth, but the root system will continue to sprout new shoots until all energy reserves are exhausted. Relying solely on cutting requires frequent repetition of treatments throughout the growing season. Applying a systemic

(top) Stem, foliage, and young flowers of oriental bittersweet in early May. (middle) Leaves and young green berries in late June. (above) Infestation climbing into canopy. Photos by Adam Gundlach, Wildlife Habitat Council

Oriental bittersweet mature berries. Photo by James R. Allison, Georgia Department of Natural Resources, Bugwood.org.

herbicide, such as glyphosate, to the cut stumps kills the root system and reduces root suckering. For larger patches where damage to surrounding vegetation is not a concern, foliar herbicide applications are most effective at reducing an infestation. Young infestations may also be dug using a weed wrench or similar tool to remove the entire root system and above ground growth.

NOTE: Oriental bittersweet has a similar appearance to American bittersweet (*Celastrus scandens*), which produces large flower (and fruit) clusters in terminal panicles at the tips of stems. Oriental bittersweet produces numerous smaller clusters from many leaf axils along the stems. American bittersweet leaves are twice as long as they are wide and tapered at each end. Oriental bittersweet leaves are smaller and generally rounded.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and Southeast Exotic Pest Plant Council Plant Manual.

Non-native swallow-worts:
Black swallow-wort (*Cynanchum louisiae*)
Pale Swallow-wort (*Cynanchum rossicum*)

Description & Biology – Black swallow-wort and pale (or European) swallow-wort are aggressive vines that still have relatively limited distributions in the Delaware River Basin and are thus high priority for control efforts, as it may be possible to eradicate or at least contain these plants before they become widespread. Control is particularly important because these species have the potential to directly impact native wildlife. They are often found overrunning fields and other open areas, and by dramatically changing the plant community structure, they have the potential to affect ground-nesting bird populations. Arthropod diversity in invaded sites is greatly diminished as compared to similar uninavded habitats. It also appears that swallow-worts can disrupt the oviposition of monarch butterflies by crowding out the monarchs' native host plants (milkweeds, which are in the same family), causing either a cessation of oviposition or an "oviposition sink" in which eggs are laid on swallow-worts rather than milkweeds and the larvae survival rate is extremely low. Researchers suspect that the non-native swallow-worts may be allelopathic (producing chemicals that inhibit growth of other plants), which would explain the ease with which they overrun habitats and crowd out native plants.

(top) Black swallow-wort flowers. (middle) Pale swallow-wort. (above) Opening seed pod. Photos by Leslie J. Mehrhoff, University of Connecticut, Bugwood.org

Swallow-worts are found in a variety of upland habitats. In addition to invading fields and meadows, they take advantage of sunlight in forest gaps to create monocultures in openings. They tolerate a wide variety of light conditions and can spread into the understory to carpet the forest floor, even though they may not flower in low light conditions.

Both species grow as herbaceous vines, though pale swallow-wort may be found growing as a forb as well. Flowers are ¼-inch-wide, borne in clusters of six to 10, and have five triangular petals that give each flower a distinctive star shape. Black swallow-wort has dark purple to almost black flowers; pale swallow-wort has creamy pink to reddish brown flowers. The native honeyvine (*Cynanchum laeve*) has white flowers. Other identifying features include a twining growth pattern and shiny oval leaves with pointed tips and entire margins. Leaves are 2 to 5 inches long by 0.5 to 3 inches wide and arranged oppositely.

Both species are perennial; black swallow-wort spreads vegetatively through rhizomes and also reproduces by seed, while pale swallow-wort lacks rhizomes and spreads primarily by seed. The flowers of both species are self-pollinating. Green tapered pods emerge in mid summer and turn light brown as they mature. When the seeds are ripe, the pods split open and seeds are dispersed by wind.

NOTE: Non-native swallow-worts may be confused with the native *Cynanchum* species honeyvine (*Cynanchum laeve*). The native species has white flowers and the base of its leaves is heart-shaped. Milkweed plants may also be confused with non-native swallow-worts, as they are in the same family and share some charactersitcs. Milkweed plants have similar seed pods, but swallow-wort pods are longer, narrower, and smoother.

Control – Non-native swallow-worts have a seed bank that will persist for up to five years. As always, it is therefore best to monitor habitat areas carefully so that new invasions can be detected early and removed before a seed bank forms. For established populations, control requires patience and repeated effort. **For any of the following control methods, it is important that any plants with seed pods (even if the plants are sprayed with herbicide) are securely bagged and discarded.**

Mechanical methods may be useful in preventing seed production to control long-distance spread, and it is possible that mowing or cutting could eventually exhaust underground reserves to the point that plant death occurs, but researchers have mainly found that these techniques do little to reduce density or percent cover in invaded areas. If mechanical control is the only suitable option, the best time to cut or mow is when immature pods are on plants (*after* flowering – plants cut during flowering can recover to produce viable seed later in the season). If digging is the desired method, the entire root crown should be removed before seeds ripen, and pods should be bagged. In cultivated areas, it may help to plant an annual crop until the seed bank is depleted (up to five years).

To control by chemical means, systemic triclopyr ester (e.g. Garlon 4) or glyphosate (e.g. Roundup Pro, Accord) can be applied as foliar spray or with a sponge. The chemical should be applied after flowering has begun, while plants are actively growing. One to two weeks after herbicide application, dead tissue spots and yellowish color should appear on the leaves, indicating that the chemical is working and the plants do not need to be sprayed again that year. Spraying will need to be repeated the following year. For cut stem application, plants should be cut low and immediately painted with glyphosate (Roundup Pro 50% solution appears to be effective).

Compiled from the Catskill Regional Invasive Species Partnership High Threat Invasive Plant Species list, Troy Weldy (Director of Ecological Management, The Nature Conservancy Eastern New York Chapter, personal communication February 2009).PCA Alien Plant Working Group fact sheets for black swallow-wort and pale swallow-wort, the University of Maine Cooperative Extension Bulletin, Averill et al. 2008, Casagrande & Dacey 2007, DiTommaso & Losey 2003 and Matilla & Otis 2003.

Chinese yam (*Dioscorea oppositifolia*)


Bulbil arising from leaf axil. Photo by Jack Ranney, University of Tennessee, Bugwood.org

Description & Biology – Chinese yam, also known as cinnamon vine, is a perennial, deciduous, twining vine native to China. It exhibits both creeping and climbing growth, reaching up to 20 feet in height as it climbs up surrounding vegetation. Vines twine from left to right. Its leaves, which generally grow to about four inches long, have a heart-shaped base with distinct leaf veins that taper to a pointed tip. The leaves can be arranged opposite or alternate and may exhibit a purplish or red coloration along leaf margins and petioles. Female flowers are borne in leaf axils as small, yellowish-white bells, while the male flowers are borne in terminal clusters at the ends of branches. Chinese yam produces seeds in a three-angled, membranous capsule, but seed viability has not been verified in the United States. Its main form of reproduction is through aerial tubers (called bulbils) that develop in leaf axils from late summer into the fall. The bulbils drop from the plant upon maturity and are dispersed in the immediate vicinity of the plant. Further long-distance dispersal is aided by wildlife and water. Bulbils are covered in adventitious buds that sprout to form new plants, even when bulbils have been partially eaten or fragmented. Bulbils are an important means of dispersal for the species and appear to be the primary means of spread over geographical areas (Mueller et al. 2003). Chinese yam has a large, tuberous root system that sprouts annually and readily sprouts after being cut or damaged.

Chinese yam is a fast-growing species that can quickly invade natural areas to form dense monospecific infestations. It displaces native vegetation by smothering and dominating sunlight, resulting in lower species richness and abundance. It typically inhabits riparian areas with rich soil, forests, roadsides, old homesteads, and waste places. It tolerates semi-dry locations and shaded


(above left) Chinese yam foliage. (above right) Climbing growth of Chinese yam up golden bamboo culms. Photos by Adam Gundlach, Wildlife Habitat Council

areas, but it rarely grows in dense shade.

Control – Established populations are difficult to eradicate. Early identification and control of Chinese yam invasions increases the likelihood of success. Small initial populations can be cut or mowed repeatedly until root reserves are exhausted, or the entire plant, including the roots, can be dug using a pulaski, weed wrench, or similar digging tool. Digging is labor- and time- intensive, and any root fragments remaining in the soil may sprout to form new plants. Large infestations are most efficiently controlled through application of systemic foliar herbicides, such as glyphosate (e.g. Accord, Roundup Pro) or triclopyr (e.g. Garlon), when leaves have fully developed but before bulbils ripen in mid to late summer. All treatment methods need to be monitored and repeated to remove new growth arising from bulbils and roots.

> **NOTE:** Chinese yam may resemble the native wild yam (*Dioscorea villosa*), but the native variety twines in the opposite direction (from right to left) and lacks aerial tubers.

Compiled from the U.S. Forest Service Weed of the Week, The Nature Conservancy Element Stewardship Abstract, Southeast Exotic Pest Plant Council Plant Manual, and Mueller et al. 2003.

Winter creeper (*Euonymus fortunei*)

Description & Biology – Also known as creeping euonymus, winter creeper is a woody, perennial evergreen vine of the bittersweet family (Celastraceae) that was introduced from China as an ornamental ground cover. As a ground cover, it can form dense, shrubby growth up to three feet tall. It also climbs trees up to seventy feet tall, attaching to the surface via aerial roots. The thick, glossy leaves are dark green or a combination of white and green, with distinct silvery veins. They are egg-shaped and occur in opposite pairs along the stem. Young twigs are smooth and lime green, slowly developing grayish streaks as they mature. Old stems are covered with corky gray bark that becomes cracked over time. Aerial roots arise from the stems and allow the vines to attach to surfaces. Winter creeper blooms from May to July, producing small greenish-yellow flower clusters at the end of y-shaped stalks. Fruits and seeds develop in the fall, beginning as reddish capsules, which split to expose seeds covered by a fleshy, reddish-orange casing, which are eaten and dispersed by birds and other wildlife. Winter creeper also spreads vegetatively through lateral shoots extending from its main branches and by new plants that develop from rootlets at short intervals.

(top) Winter creeper foliage. (middle) Dense winter creeper growth as a landscaping ground cover. (above) Winter creeper flowering. Photos by Adam Gundlach, Wildlife Habitat Council.

Winter creeper proliferates in a variety of less-than-optimal conditions including poor soils, dense shade, and a range of pH values, but it does not grow well in heavy, saturated soil. Its ability to reproduce vegetatively from lateral shoots and rootlets allows it to quickly form dense infestations that displace native vegetation by dominating sunlight, nutrients, water, and space. Vines growing up trees can reduce the trees' photosynthetic capabilities and increase the risk of blow downs during high winds.

Control – For small initial populations or locations where herbicide is not an option, the entire plant including the root system can be removed using a pulaski, weed wrench, or similar tool, though this method is time- and labor-intensive. Immature plants with poorly developed roots can be pulled by hand. All vegetative material should be bagged and properly disposed. Any root fragments remaining in the soil may sprout to form new plants. Large infestations can be treated with cut-stem or foliar applications of glyphosate (e.g. Roundup Pro, Accord) or triclopyr (e.g. Garlon) herbicides. The cut-stem method is more selective than foliar application and thus better for areas where damage to non-target vegetation is a concern.

Compiled from the U.S. Forest Service Weed of the Week, PCA Alien Plant Working Group Fact Sheet, and Plant Invaders of Mid-Atlantic Natural Areas.

English ivy (*Hedera helix*)

English ivy leaves. Photo by Adam Gundlach, Wildlife Habitat Council.

Description & Biology – English ivy is a highly invasive evergreen vine that was likely introduced to the United States by early European immigrants. Today, it is still widely sold as an ornamental ground cover due to its hardy, evergreen growth. English ivy has a vigorous growth habit that quickly spreads to cover any surface in its path, attaching by tiny root-like structures that arise from the stem and exude an adhesive substance. The dark green, waxy leaves alternate along the stem and are typically three-lobed but can exhibit great variability – round leaves lacking lobes are often found on plants ready to flower. In locations that receive adequate sunlight, English ivy produces flowers on stalks that extend out from the main vine at right angles. Flowers are borne as small, white to greenish, umbrella-like clusters that bloom in the fall. The berry-like fruits are deep purple to black and contain small, stone-like seeds at maturity. English ivy reproduces through seeds and by vegetative means. Seeds are dispersed to new locations mainly by birds. Stem and root fragments separated from the parent plant can root to form new plants when in contact with soil.

English ivy is shade tolerant, can invade a variety of habitats including woodlands, fields, forest edges, hedgerows, fence lines, and other waste places, and invades all levels of vegetation from understory to canopy. As a dense, creeping ground cover, it excludes native forbs and tree seedlings by preventing sunlight from reaching the soil. As vines grow up tree trunks, they slowly engulf the entire host tree, first covering the trunk, then lower branches, and eventually canopy branches, which ultimately results in death of the tree. The added weight in the crown of trees increases the risk of blow downs during high winds. In addition, English ivy is a known reservoir for bacterial leaf scorch (*Xylella fastidiosa*), which is a plant pathogen that affects native trees such as elms, oaks, and maples.

(above left) English ivy berries. (above right) Young vine with roots. Photos by Adam Gundlach, Wildlife Habitat Council.


English ivy engulfing the trunks of two trees. Photo by Adam Gundlach, Wildlife Habitat Council.

Control – Small patches of English ivy can be hand pulled. Effort should be made to remove the entire root system. All plant fragments should be bagged and disposed, as any fragments left in the soil may root to form new plants. Vines growing up trees can be cut off near the base to kill the upper growth and reduce stress on the canopy. Cut stems can be treated with systemic herbicide, such as glyphosate or triclopyr, to kill remaining underground portions of the plant. Systemic herbicide may also be applied to extensive infestations as a foliar application or basal bark treatment. Monitoring and repeated control treatments will likely be needed in subsequent years to ensure complete control.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and Plant Invaders of Mid-Atlantic Natural Areas.

Japanese hop (*Humulus japonicus*)

Description & Biology – Japanse hop is an annual vine in the hemp family (Cannabaceae) that is closely related to *Humulus lupulus*, the hop species often used in beer brewing. Its growth habit varies from trailing to climbing to twining to prostrate. Its leaves are two to four inches across, palmately lobed with five lobes, and have a rough surface. Bracts develop at the base of leaf petioles near the stem and are down-curved, a distinguishing feature for this species. The bright green stems are covered with hooked prickles that cause discomfort during hand pulling of plants unless proper gloves are used. Inconspicuous green flowers bloom in August and September, with male and female flowers on separate plants. Japanese hop produces small seeds that can be dispersed by wind and water movement. It typically regenerates each year from seed but occasionally may exhibit perennial growth in certain areas.

Japanese hop grows in moist soils and is generally found in abandoned fields, along roadsides and forest edges, and near stream banks. It tolerates various soil conditions from sandy to clay-dominated soils and from acidic to alkaline pH soils. In well-established infestations, the quick growth rate of Japanese hop can easily smother and displace native vegetation.

Control – Small outbreaks can be removed manually by hand pulling and digging to remove as much of the root system as possible. Removal should occur in August or September prior to development of seeds, and sturdy gloves should be worn due to the prickly stems of Japanese hop. Most general-use herbicides, such as glyphosate, applied as a foliar spray effectively control large infestations. The seedbank should be exhausted after two or three years of control.

(top) Japanese hop foliage. (middle) Young leaves with developing flower bud. (above) Japanese hop infestation on a compost pile at Defense Supply Center Richmond. Photos by Adam Gundlach, Wildlife Habitat Council.

NOTE: Wild cucumber (*Echinocystis lobata*) also has palmately five-lobed leaves and similar growth but produces tendrils and lacks prickles on the stem.

Compiled from the U.S. Forest Service Weed of the Week, The Nature Conservancy Weed Notes, and Invasive Plant Atlas of New England.

Japanese honeysuckle (*Lonicera japonica*)

(top) Japanese honeysuckle flowering. (above) Young vine with typical opposite leaves. Photos by Adam Gundlach, Wildlife Habitat Council.

Description & Biology – Japanese honeysuckle is a perennial woody vine native to Japan and Korea that was introduced to the United States in the 1800's for a variety of purposes. Its opposite leaves can be broadly oval-shaped to elliptic with blunt or rounded tips and short stalks. Populations in southern or mid-Atlantic regions often remain evergreen through the winter, while northern populations generally drop their leaves following sustained cold temperatures. Stems are slender, light brown or reddish, and somewhat pubescent (hairy) when young. Mature stems can reach up to 80 feet long and are smooth, hollow and covered in brown bark that peels in strips. Japanese honeysuckle produces fragrant, white, tubular flowers (composed of five fused petals) in pairs at leaf nodes. Flowers bloom from April through July, sometimes extending as late as October. As flowers reach senescence, they take on a yellow coloration distinct from young white flowers. In the fall, small, black, berry-like fruits develop, with each fruit containing two to three dark brown, oval seeds. Japanese honeysuckle reproduces both sexually through seeds and vegetatively through rhizomes and runners (stolons), which root at any leaf nodes that contact moist soil.

Japanese honeysuckle has a vigorous trailing to climbing growth habit that can girdle and smother shrubs and small trees. It typically invades fields, forest edges and openings, wetlands, floodplains, and disturbed habitats. Its semi-evergreen to evergreen growth provides a nearly year-round growing season that gives it a competitive advantage over native species. Dense infestations can kill other plants by preventing sunlight from reaching their leaves, and the vigorous root system allows Japanese honeysuckle to spread quickly and dominate soil nutrients and water.

Control – Small patches of Japanese honeysuckle can be eliminated by repeated pulling of vines and as much of the root system as possible. Frequent monitoring and removal of new growth ensure successful control. Cutting and pulling above ground growth weakens the plant but does not kill it, as new shoots develop from roots and any cuttings left in contact with soil. Mowing is only effective if performed several times per year for several years. Dense infestations can be controlled with foliar applications of herbicide such as glyphosate or triclopyr. Optimal treatment time is in the fall when most native non-target plants are entering dormancy and Japanese honeysuckle is still actively growing. Herbicide applications can continue throughout the winter in locations where temperatures remain above the required minimum, as outlined on the herbicide label.

Combinations of treatments often show the best results. Mowing an infested area and then applying herbicide once new growth develops from the root system increases effectiveness of control. Another method involves burning an infestation in the fall to remove accumulated biomass and then in the spring applying herbicide once new growth has emerged.

(top) Lobed form of young leaves. (above) Mature Japanese honeysuckle vines next to trunk of host tree. Photos by Adam Gundlach, Wildlife Habitat Council.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, and The Nature Conservancy Element Stewardship Abstract.

Mile-a-minute (*Polygonum perfoliatum*)

Mile-a-minute stems and typical triangular leaves. Photo by Britt Slattery, U.S. Fish and Wildlife Service, Bugwood.org

Description & Biology – Mile-a-minute is an annual, herbaceous, trailing vine native to southern and eastern Asia. It is a new invader in many portions of the Delaware River Basin (especially the northern areas in New York) and is thus a priority for immediate control of any populations that appear. Its slender stems are green when young, turning reddish with age, and are covered in tiny downward curved barbs. The leaves are distinctly triangle-shaped with barbs on the underside and an alternate arrangement along stems. Stems are also surrounded at intervals by circular, funnel-shaped leaf-like structures called ocreae. The inconspicuous white flowers arise from the ocreae, blooming from June through early fall. Metallic blue, segmented fruits are produced, with each segment containing a small, glossy, black seed. Seeds can be transported great distances by water, as they remain buoyant for many days. Birds facilitate long-distance dispersal, while certain species of ants have been observed to transport seeds locally.

Mile-a-minute flower surrounded by the ocreae. Photo by Jil M. Swearingen, USDI National Park Service, Bugwood.org

Mile-a-minute favors areas with moist soil and tends to invade open disturbed sites, including fields, riparian areas, forest edges and openings, wetlands, and roadsides. It requires sunlight for the majority of the day and only tolerates partial shade. It grows rapidly and, using its barbed stems, can climb over other vegetation to reach sunlight. Dense infestations prevent sunlight from reaching the smothered plants below and prevent germination and seedling development of native species, leading to reduced biodiversity.

Control – Mile-a-minute vines can be easily pulled by hand. Gloves should be worn when pulling mature vines, as they have hardened, recurved barbs. Plants that have already set seed should not be pulled, as this action may distribute seeds to new locations. Repeated cutting or mowing prevents seed production and if continued for several years eventually exhausts the seedbank. Various general-use herbicides are effective at controlling extensive infestations; however, mile-a-minute is not listed on most product labels and herbicide treatments require approval by the site's State Department of Agriculture.

Compiled from the PCA Alien Plant Working Group Fact Sheet, Plant Invaders of Mid-Atlantic Natural Areas, Van Driesche et al. 2002, and Troy Weldy (Director of Ecological Management, The Nature Conservancy Eastern New York Chapter, personal communication February 2009).

Kudzu (*Pueraria montana*)

Trifoliate kudzu leaves. Photo by Adam Gundlach, Wildlife Habitat Council.

Description & Biology – Kudzu is an aggressive, perennial, semi-woody vine of the pea family (Fabaceae) that is native to Asia. It was widely planted in the southeastern United States in the mid-1900s for soil erosion control in agricultural areas, and by 1970 it had been listed as a common weed in the south. It has deciduous leaves that drop from the plant after the first frost. Each leaf is composed of three leaflets, which can be up to four inches in diameter. Leaflets have hairy margins and their shape varies from entire to multi-lobed, usually two or three lobes. Stems are hairy and pliable when young, becoming fibrous as they mature, and can grow up to four inches in diameter. The climbing vines can reach as much as 100 feet long, growing one foot per day under optimal conditions and 60 feet in a single growing season. The fragrant, purple flowers are borne in long clusters (called racemes) and bloom from July through October. The flowers give way to hairy, two-inch long pods containing three or more hard seeds. Currently in the United States, kudzu reproduces almost solely by vegetative means from rhizomes, runners, and vines, which root at nodes when contacting soil to form new plants. A small percentage of seeds produced by kudzu are viable, but they generally do not germinate for several years. Its root system is extensive and hardy, producing a taproot that can be more than six feet long, seven inches in diameter, and weigh hundreds of pounds. As many as thirty vines can arise from a single root crown. When vines root at nodes, the nodes form new root crowns and taproots, which in turn can produce more vines.

(above left) Kudzu seedpods. Photo by Ted Bodner, Southern Weed Science Society, Bugwood.org. (above right) Kudzu flower cluster. Photo by Chuck Bargeron, University of Georgia, Bugwood.org.

Kudzu typically grows in moderate climates that experience mild winters, warm summers, and abundant rainfall, but it survives in a variety of environmental conditions and has recently been observed to survive farther north than previously believed. It can generally be found growing in forest edges, roadsides, fields, and a variety of other open disturbed areas. It also can persist in dense shade of the forest floor until climbing up trees toward the light of canopy gaps. The large root system allows established populations to survive drought conditions. Kudzu's vigorous growth forms a dense blanket that smothers existing vegetation, including trees, which are engulfed by the vines, creating an almost topiary effect in extensive infestations. The added weight of vines often snaps tree branches or uproots entire trees. Kudzu infestations significantly alter plant communities by smothering existing vegetation and preventing germination and seedling development of native plants, leading to reduced biodiversity.

Control – To successfully control kudzu infestations, its persistent root system must be destroyed. Cutting or mowing monthly for two or more growing seasons may achieve control. All vegetative material should be disposed of by feeding to livestock, burning, or bagging. Late season cutting followed by an immediate application of systemic herbicide will promote uptake to the roots. A variety of herbicides – glyphosate, triclopyr, picloram, and metsulfuron – can be used for foliar applications. Certain persistent, soil-active herbicides (Imazapyr) have proven effective at reducing large infestations in forestry management. Treatment areas need to be frequently monitored to ensure that all root crowns have died, as only a single surviving root crown could spread to form a new infestation.

Kudzu infestation on a bluff overlooking the Chesapeake Bay, mid August. Photo by Adam Gundlach, Wildlife Habitat Council.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, Southeast Exotic Pest Plant Council Plant Manual, and Van Driesche et al. 2002.

Periwinkle (*Vinca major & V.minor*)

Description & Biology – Periwinkle, or vinca, is a common sight in gardens and is readily available for sale at plant nurseries. Allegedly sterile cultivars can even be found at "native" plant nurseries. Unfortunately, whether its seeds are viable or not, this trailing vine spreads vegetatively by rooting at nodes and forms dense infestations in forest understories, where it alters forest dynamics significantly by inhibiting tree regeneration. Periwinkle may also spread via waterways. These species often grow in riparian areas, and there are reports that plant parts moved downstream by the current may be capable of starting new infestations.

Both common periwinkle (*Vinca minor*) and bigleaf periwinkle (*Vinca major*) were introduced from Europe in the 1700s. They are often found spreading in dense mats from old homesites into natural areas. A single clone can overtake a large area of woodland understory, crowding out the native understory species.

Periwinkle vine with characteristic pinwheel-shaped flowers. Photo by Dan Tenaglia, Missouriplants.com, Bugwood.org

Periwinkles are evergreen to semi-evergreen vines, succulent when young and becoming somewhat woody with age. They creep along the ground; flowering branches stand up straight. Their flowers have a pinwheel shape and are usually violet, though blue and white varieties exist as well. The glossy, hairless leaves occur in pairs along the stems and range from elliptic to heart-shaped. Leaf color is usually dark green with distinct light-colored midvein. Leaves may also be variegated (green with white edges). Slender, cylindrical fruits emerge in late spring to early summer.

Control – Elimination of these species from the nursery trade and gardening circles would be a major step forward. Established plants can be removed with a rake or three-pronged fork when soil is moist. Roots must be carefully extracted from the ground at each node, with care taken to avoid leaving root fragments in the soil. Cutting in early to late spring followed by application of glyphosate (e.g. Roundup Pro, Accord) can also be effective. Foliar spray of glyphosate or triclopyr (Garlon 4 2%) is effective when applied during warm days in fall or winter for successive years. Resprouts should be carefully removed with roots intact or spot-treated with herbicide.

NOTE: The native partridgeberry (*Mitchella repens*) also has pairs of glossy dark leaves and creeps along the ground. It can be distinguished from vinca by its hairy white four-petaled flowers, scarlet berries and ovate to cordate leaves.

Compiled from: Plant Right, Lady Bird Johnson Native Plant Information Network, J. H. Miller's Nonnative invasive plants of southern forests: a field guide for identification and control, and Darcy & Burkart 2002.

Exotic Wisterias:
Japanese wisteria (*Wisteria floribunda*)
Chinese wisteria (*Wisteria sinensis*)

Description & Biology – Chinese and Japanese wisteria are perennial – sometimes living up to 50 years – woody vines in the pea family (Fabaceae) that were introduced to the United States as ornamental species for their showy flower clusters and foliage. Leaves are alternate, pinnately compound, and grow to nearly one foot in length. Japanese wisteria leaves are comprised of 13 to 19 smaller leaflets, while Chinese wisteria leaves are comprised of 7 to 13 leaflets. Stems of Japanese wisteria are white-barked and twine clockwise, while Chinese wisteria stems are dark gray and twine counter-clockwise. Vines can reach ten inches in diameter and have densely hairy twigs. Flowers are borne in long, showy clusters (racemes) that hang from vines. Flowers bloom from March to May and vary in color from purple to bluish or even pink. Seed pods mature from July to November, are four to six inches long, covered in velvety brown hair, and contain up to eight brown, flattened, round seeds. American wisteria (*Wisteria frustescens*), produces smaller seed pods that lack velvety hairs. The main form of reproduction is through abundant stolons, which readily produce new roots and shoots. Seeds can be transported by water and human activity to new colonize new locations.

(above left) Chinese wisteria flower cluster. Photo by Chris Evans, River to River CWMA, Bugwood.org. (above middle) Wisteria compound leaf and stem. (above right) Wisteria seed pod. Photos by Ted Bodner, Southern Weed Science Society, Bugwood.org.

Exotic wisterias generally grow in locations that receive full sun, though established plants can tolerate partial shade. They also tolerate a variety of soil and moisture combinations and can typically be found growing in forest edges, roadsides, and rights-of-way. As vines grow and cover surrounding vegetation, the twining vines wrap tightly around stems, strangling small trees and shrubs and preventing other vegetation from growing. Vines often grow into the canopy of trees, forming dense growth that can eventually kill the host tree through shading and strangling.

Chinese wisteria infestation creeping up pine trees. Photo by Chris Evans, River to River CWMA, Bugwood.org

Control – For small populations and areas where herbicide use is not feasible, vines can be cut at the root collar to kill above ground growth and relieve stress on host trees. Cutting alone will require repeated treatments to eliminate resprouts. The root system can be dug using a weed wrench or similar tool to prevent new sprouts from developing. All vegetative material should be bagged and removed from the site. Any root fragments left in the soil can sprout to form new plants. To increase efficacy, a systemic herbicide can be applied directly to stumps immediately following cutting in order to kill the root system and avoid digging. For large infestations where damage to surrounding vegetation is not a concern, a foliar application of systemic herbicide may be feasible depending on the height of vines. Vines that have grown high into the canopy should first be cut, allowed to resprout, and then treated with a foliar herbicide application after sufficient regeneration has occurred.

NOTE: Two native *Wisteria* species occur in the southeastern United States. Native and non-native species are similar in appearance, but the introduced species exhibit hardier, more aggressive growth and more fragrant flowers compared to native species. Positively identify the species before beginning control measures.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, Plant Invaders of Mid-Atlantic Natural Areas, and Southeast Exotic Pest Plant Council Plant Manual.

INVASIVE TREES & SHRUBS

COMMON NAME	SCIENTIFIC NAME	PAGE
Norway maple	*Acer platanoides*	70
Tree-of-heaven	*Ailanthus altissima*	72
Mimosa	*Albizia julibrissin*	74
Japanese barberry	*Berberis thunbergii*	76
Paper mulberry	*Broussonetia papyrifera*	78
Russian olive	*Elaeagnus angustifolia*	79
Autumn olive	*Elaeagnus umbellatus*	81
Winged burning bush	*Euonymus alatus*	83
Privets	*Ligustrum* spp.	85
Bush honeysuckles	*Lonicera* spp.	87
Chinaberry tree	*Melia azedarach*	89
White mulberry	*Morus alba*	91
Princess tree	*Paulownia tomentosa*	92
Amur corktree	*Phellodendron amurense*	94
White poplar	*Populus alba*	96
Bradford (Callery) pear	*Pyrus calleryana*	97
Common buckthorn	*Rhamnus cathartica*	98
Black locust	*Robinia pseudoacacia*	100
Multiflora rose	*Rosa multiflora*	102
Wineberry	*Rubus phoenicolasius*	104
Japanese spiraea	*Spiraea japonica*	105
Siberian elm	*Ulmus pumila*	106

Adapted from PCA Alien Plant Working Group – Mid-Atlantic List

Norway maple (*Acer platanoides*)

Description & Biology – Norway maple is a deciduous tree in the maple family (Aceraceae) that was introduced from Europe as an ornamental plant for landscaping. It typically reaches heights of 40 to 60 feet but can grow up to 100 feet tall, with a broad, rounded crown. The large leaves are four to seven inches across, glossy, and dark green, and the leaf veins and petioles contain a milky substance, which native maples do not contain. Leaves are palmate, comprised of five to seven lobes, with numerous sharply-tipped points around the leaf margin, and occur in opposite pairs along stems. The bark in young plants is smooth and gray becoming darker and furrowed as the tree matures. Bright yellow-green flower clusters appear between April and May prior to the appearance of leaves. During summer, the fruits mature into pairs of winged samaras fused at their base. Each samara contains a single seed and the pair later splits apart and glides to the ground in a helicopter-like fashion. Norway maple produces prolific amounts of seed that readily germinate and crowd out other species near the parent plant. Additionally, it spreads vegetatively from the roots, further adding to local infestations.

(top) Norway maple foliage. (middle) Young flowers emerging from bud. (above) Bark of trunk. Photos by Paul Wray, Iowa State University, Bugwood.org.

Norway maple has escaped cultivation and can now be found invading natural areas, including forests, fields, and other habitats. It can become a dominant species in many locations, displacing native trees and shrubs, while the dense foliage shades out native forbs.

Control – Seedlings can be pulled by hand when soil is moist. Saplings can be dug, removing as much of the root system as possible. Mature trees can be cut down; repeating as necessary to remove resprouts from the stump. Systemic herbicides such as triclopyr and glyphosate have also proven effective at control. Herbicide can be applied as a foliar application to young trees. Mature trees can be treated with hack-and-squirt, basal bark, or cut stump treatments. In natural areas, trees can be girdled and left standing to create snags (standing dead trees) for cavity-nesting species. Sprouts that develop following girdling should be removed.

Compiled from the U.S. Forest Service Weed of the Week, and Plant Invaders of Mid-Atlantic Natural Areas.

Tree-of-heaven (*Ailanthus altissima*)

Tree-of-heaven leaflets and samaras. Photo by Adam Gundlach, Wildlife Habitat Council.

Description & Biology – Tree-of-heaven, also called Chinese sumac, stinking sumac and paradise tree, is a fast-growing, deciduous tree native to central China. It is typically associated with tropical or subtropical climates in its native range but has proven to be tolerant of a variety of conditions in the United States. Most of the plant parts, especially the flowers, have a pungent odor. Its foliage is similar to that of native sumac (*Rhus* spp.), and can easily be mistaken without proper identification. The alternate leaves are pinnately compound, comprised of 11 to 25 or more leaflets, and can grow to four feet in length. Leaflets are lanceolate, one to two inches wide and three to five inches long, and have a pair of gland-like nodes at their base. The leaf scars that remain on branches after leaves drop in the fall are heart-shaped to triangular, a characteristic that distinguishes tree-of-heaven from native sumac species. Trees can reach more than 80 feet tall with smooth, gray bark that becomes cracked with age. It is a dioecious species with male and female flowers occurring on separate trees, though both may occasionally be found on a single tree. The numerous yellow-green flowers bloom from May through June. Seeds develop in late summer on female trees as pink, papery samaras, which remain on the tree into winter as pink clusters. Tree-of-heaven reproduces both through seed and vegetatively through root sprouts, especially following injury. True seedlings are typically smaller than root sprouts, as they have a smaller reserve from which to draw energy. In established colonies, reproduction is primarily through root sprouts.

Tree-of-heaven leaves and samara cluster. Photo by Adam Gundlach, Wildlife Habitat Council.

Tree-of-heaven is shade intolerant but tolerates a variety of adverse conditions (pollution, drought, poor soil) and can be found growing in most any open, disturbed habitat, such as roadsides, fencerows, and fields. In urban areas, it is often observed growing through cracks in pavement. Its vigorous growth can quickly form dense thickets, which displace native vegetation. In addition, it releases allelopathic chemicals, which prevent other plants from growing in its vicinity.

Tree-of-heaven. Photo by Adam Gundlach, Wildlife Habitat Council.

Control – Control requires patience and repeated treatments, as tree-of-heaven sprouts profusely from the root system when damaged. Young seedlings can be hand pulled, preferably when the soil is moist; removing the entire root with the seedling to prevent resprouts. Cutting down large, seed-producing female trees can help decrease the appearance of seedlings, as a single tree can produce thousands of seeds each growing season. Following cutting, abundant root sprouts appear in the area surrounding the cut tree and require repeated removal until energy stores are exhausted. To reduce resprouting, cut stumps can be treated with herbicide to decrease vigor of the root stock.

In locations where feasible, herbicide application is the most effective method for controlling large infestations. The appropriate type of herbicide to use depends on the specific site conditions. Glyphosate can be used as a foliar spray in areas where damage to non-target plants is not a concern. Triclopyr, a broadleaf herbicide, can be used in areas where desirable grasses are growing in association with tree-of-heaven. For locations where a more directed application is required, herbicide can be applied via the basal bark method or the hack-and-squirt method.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and Southeast Exotic Pest Plant Council Plant Manual.

Mimosa (*Albizia julibrissin*)

Description & Biology – Also known as silktree and silky acacia, mimosa is a deciduous tree that is native from Iran to China and Japan. It was introduced to the United States as an ornamental for its feathery foliage and showy, fragrant flowers. Mimosa is a shortlived small to medium tree, growing from 20 to 40 feet tall, with smooth, gray bark. The leaves are highly segmented (bipinnately compound), fern-like, and composed of 10 to 25 pinnae, each with 40 to 60 asymmetric leaflets. The entire leaf structure is from five to eight inches long and three to four inches wide. Showy pink flower clusters develop at the end of branches, blooming from May through August, and resemble pink pom-poms. The flowers give off a strong, sweet fragrance. Seeds are produced in flattened, light brown seed pods that develop from August through September. The six-inch long seed pods persist on trees into the winter months, eventually breaking apart to reveal the light brown, oval seeds, which have a thick seed coat that allows them to remain dormant and viable for many years. Mimosa reproduces mainly by seed, but will resprout soon after being cut or damage.

(top) Mimosa foliage and flower. (above) Flowering tree. Photos by Adam Gundlach, Wildlife Habitat Council.

Mimosa is a moderate threat to natural areas and is often found growing in forest edges, along roadsides, and in other areas of disturbance; it also is commonly found in riparian areas where seeds are transported by water and germinate on scoured stream banks. Though tolerant of partial shade, it typically grows in locations with full sun. Its ability to grow in a variety of soils and produce abundant, persistent seed crops allows it to be highly competitive with native vegetation. Dense stands of mimosa reduce the sunlight and nutrients available to native plants.


Developing mimosa seed pods. Photo by Adam Gundlach, Wildlife Habitat Council.

Control – Mimosa can be controlled with mechanical and chemical techniques. Trees may be cut down as an initial control, but will require repeated cutting or other control methods to eliminate resprouts. Applying a systemic herbicide to the cut stump immediately after cutting will inhibit resprouts from forming. Large trees can be girdled to kill above ground growth where herbicide use is not feasible, but will also require repeated control of resprouts. Young seedlings can be hand pulled once they have reached an adequate size, making sure to remove as much of the root as possible. Application of systemic herbicide will kill the root system and prevent resprouts. Herbicide can be applied as a foliar application to seedlings and small trees. For larger trees, a basal bark treatment or cut-stump application will likely be more successful.

Compiled from the PCA Alien Plant Working Group [Fact Sheet](), and Southeast Exotic Pest Plant Council [Plant Manual]().

Japanese barberry (*Berberis thunbergii*)

Description & Biology – Japanese barberry is a deciduous shrub introduced in the mid-1800s as an ornamental species for landscaping. Its dense, spiny growth generally reaches two to four feet in height, but can grow to eight feet. The branches are brown with three ridges or grooves and a thorn is produced at each node. The small, oval-shaped leaves vary from bright green to burgundy in coloration, are semi-evergreen, and alternate along stems, often in clusters. Pale yellow flowers develop in thorn axils in clusters of two to four, and bloom from March into May. Numerous bright red, oblong berries are borne on tiny stalks and ripen in late summer or fall, persisting on the plant through the winter. It reproduces from both vegetatively and through seed, which exhibits a high germination rate. Birds and small mammals eat the berries and disperse the seeds to new locations. Its creeping rhizome system can send up shoots to form new plants, and branches contacting the ground can root to form new plants. This allows a single plant to spread locally to form a dense thicket.

(top) Red barberry foliage and stem with thorns. Photo by Adam Gundlach, Wildlife Habitat Council. (above) Flowering barberry with green foliage. Photo by Leslie J. Mehrhoff, University of Connecticut, Bugwood.org

Japanese barberry invades a variety of habitats, including wetlands, meadows, open and closed canopy forests, and disturbed areas. It is highly shade tolerant, resists drought, and is not consumed by deer and other herbivores, which may increase its ability to invade natural areas. Its widespread use in landscaping and abundant seed production increases the likelihood of dispersal to natural areas. Infestations can shade out native understory species and may result in altered soil chemistry.

(above left) Barberry plants. Photo by Adam Gundlach, Wildlife Habitat Council. (above right) Barberry fruits. Photo by Barry Rice, sarracenia.com, Bugwood.org.

Control - Japanese barberry leafs out prior to most native species in the spring, making it easy to identify and distinguish during this time period. Small infestations can be controlled by digging or hand pulling plants, including all roots, when the soil is moist and loose; its shallow root system makes this relatively easy. Use thick gloves to protect hands from the thorny stems. For extensive infestations that negate mechanical or manual removal, systemic herbicides, such as glyphosate or triclopyr, have proven effective at control. These herbicides can be applied as foliar, cut-stump, or basal bark applications. Performing herbicide treatment early in the spring will limit negative affects to native vegetation.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, and Southeast Exotic Pest Plant Council Plant Manual.

Paper mulberry (*Broussonetia papyrifera*)

Description & Biology – Paper mulberry is a deciduous tree native to Japan and Taiwan that has been identified as an invasive weed in at least twelve countries around the world. It is a small to medium tree, growing to a height of about 45 feet, with soft, weak wood. Its large leaves are covered in dense, fine, gray hairs, and the upper surface is rough to the touch. Leaves can be entire or multi-lobed with deep incisions, the margins are finely serrated, and leaf arrangement varies from alternate to opposite to whorled. Young stems and twigs are reddish brown and hairy, and the bark is tan or pale gray and smooth, forming shallow fissures with maturity. Paper mulberry is a dioecious species with male and female flowers produced on separate trees. Male flowers are borne in long clusters, similar to catkins of birch species. Female flower clusters are borne in unusual filamentous balls that have burgundy to purple follicles. The female flower clusters develop into ball-shaped aggregate fruits that begin green but shift to reddish purple or orange as they mature. Paper mulberry reproduces by seed and vegetatively. The fruits are eaten by wildlife, which disperse the seeds to new locations. It spreads locally by sprouting new trees from the root system. It is typically found growing in open habitats such as forest edges, fields, and other disturbed areas, and is especially apt to invade floodplains and riparian areas.

Control – Seedlings can be pulled by hand when the soil is moist. Young trees can be repeatedly cut until no new sprouts develop. Larger trees can be cut down and the stump treated with systemic herbicide to prevent resprouts from forming. Herbicide can also be applied via basal bark, hack-and-squirt, and injection methods.

(top) Various leaf forms of paper mulberry. Photo by Adam Gundlach, Wildlife Habitat Council. (middle) Male flower cluster. Photo by Gerald D. Carr, Carr Botanical Consultation, Bugwood.org. (above) Note the round aggregate fruits (arrow) developing on a female tree. Photo by J. Scott Peterson, USDA NRCS, Bugwood.org.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and U.S. Forest Service Weed of the Week.

Russian olive (*Elaeagnus angustifolia*)

Russian olive tree. Photo by Chris Evans, River to River CWMA, Bugwood.org.

Description & Biology – Russian olive is a small tree or large shrub native to Eastern Europe and Western Asia that was formerly recommended by many agencies for wildlife and windbreak plantings. It is similar in appearance to its close relative autumn olive (*Elaeagnus umbellata*), but can be distinguished by Russian olive's longer, narrower, and generally more silvery leaves (autumn olive's leaves tend to be greener) and its yellow fruits (autumn olive's generally are red or pink and juicy). Russian olive grows from 15 to 30 feet tall with a rounded, open crown. The simple leaves are oblong to lance-shaped, two to four inches long, and alternate along the stems. The upper leaf surface is light green with silvery hairs and the lower surface is covered in dense silvery white scales. The stems produce thorns and silvery to rusty scales cover the stems, buds, and leaves. After three years of growth, Russian olive is begins to produce flowers and fruit. Small, yellow flowers are borne in aromatic clusters from leaf axils. Trees typically bloom shortly after leaves emerge in early summer. Flowers develop into small olive-shaped fruits that are relatively hard and mealy. Fruits are generally light green to yellow (sometimes red) and each contains a single nutlet, which is dispersed by birds that feed on the fruit. It reproduces and spreads mainly by seed, but the root crown is capable of producing new shoots and root suckers following damage or disturbance to the above ground tree.

Russian olive is capable of growing in variety of habitats, due in large part to its nitrogen fixing ability, and has been planted extensively for erosion control and highway borders. It is most often found invading stream banks, fields, roadsides, and grasslands. It exhibits some shade tolerance, allowing it to survive in the understory of wooded areas and become dominant after the canopy species die off.

(above left) Russian olive foliage and flowers. Photo by Paul Wray, Iowa State University, Bugwood.org (above right) Russian olive fruits. Photo by Patrick Breen, Oregon State University, Bugwood.org.

Control – Identifying Russian olive early in its establishment makes control much easier. Young seedlings can be pulled by hand and saplings can be dug or removed with a weed wrench in order to remove all portions of the root to prevent resprouts. Once established, it is a difficult species to eradicate and takes many years of persistent management to control. A variety of techniques can be employed to control Russian olive including cutting, burning, mowing, digging, and herbicide application. A combination of techniques often yields the best results. All methods that remove or damage the above ground growth without damaging the root system will result in vigorous resprouting and potentially a denser infestation. Mature trees are most effectively and directly treated via cut-stump, hack-and-squirt, or girdle applications of systemic herbicide, which will kill the root system. In dense infestations, foliar and basal bark applications can also be made, but increase the risk of damage to non-target species.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, and The Nature Conservancy Element Stewardship Abstract.

Autumn olive (*Elaeagnus umbellata*)

Description & Biology – Autumn olive is deciduous shrub or small tree, growing to around 20 feet in height, which was introduced from East Asia and widely promoted for erosion control, ornamental landscaping, wildlife plantings, and wind breaks. Its green foliage, upward spreading crown, and bright red fruit distinguish it from the closely related Russian olive. The alternately arranged leaves are dark green above with silvery white scales on the underside, smooth margins, and an elliptic or ovate shape. The bark is golden brown to silver and prominent spines often develop on twigs. Autumn olive blooms from May through June, producing yellow to white, tubular flowers from leaf axils. A prodigious amount of juicy, round fruits are then produced, turning from silver as immature fruits to red with silver or brown scales as they mature in September and October. A single plant can produce hundreds of thousands of seeds in a year. The seeds have a high germination rate and are a popular food of birds, which distribute them across the landscape.

(top) Autumn olive shrub-like growth. Photo by Chris Evans, River to River CWMA, Bugwood.org. (above) Flower and leaf underside. Photo by James H. Miller, USDA Forest Service, Bugwood.org

Autumn olive begins growth early in the year, generally mid-March to early April depending on the location. Autumn olive is not tolerant of shade, but is drought tolerant. It grows well in a variety of soils, and like Russian olive, its ability to fix nitrogen via bacterial root nodes allows it to grow in poor soil conditions. It generally invades open disturbed areas, fields, forest edges, open woodlands, and roadsides.

(above left) Autumn olive fruits and foliage. Photo by James R. Allison, Georgia Department of Natural Resources, Bugwood.org. (above right) Autumn olive shrub-like growth. Photo by Chris Evans, River to River CWMA, Bugwood.org.

Control – Young seedlings can be pulled by hand. Pulling should be performed following rain when the soil is moist and loose. Mature plants are most effectively controlled by systemic herbicide via cut-stump, hack-and-squirt, or foliar applications. Prescribed burns can also be employed for control, but should be combined with other control techniques to increase efficacy. Cutting or burning without any further control will result in a denser infestation, as autumn olive vigorously resprouts from the roots following such disturbance.

Compiled from the U.S. Forest Service Weed of the Week, and The Nature Conservancy Element Stewardship Abstract, and Southeast Exotic Pest Plant Council Plant Manual.

Winged burning bush (*Euonymus alatus*)

Description & Biology – Also called winged euonymous, this deciduous shrub in the bittersweet family (Celastraceae) was introduced from China and other parts of Asia as an ornamental species. Its name derives from the thin, corky, wing-like segments that develop along the green to brown stems. Mature plants can grow from 15 to 20 feet in height and nearly as wide, but most plants are five to ten feet tall. The elliptic leaves are one to three inches long with finely serrated margins, develop in pairs along the stems, and are dark green in summer, becoming brilliant red in fall – a main reason for its use in landscaping. Inconspicuous green flowers bloom from May into early June and develop into smooth red-orange to purple fruits in fall.

Winged burning bush has been observed to invade woodlands and fields where it has escaped from cultivated landscapes. It is adaptable to a variety of conditions and is shade tolerant, but does not grow well in soils that are prone to saturation or drought. Birds and other animals disperse the seeds to new locations. Once established, it spreads locally through root suckering and seeds, forming dense thickets that crowd out native forbs and shrubs.

Control – Winged burning bush saplings up to two feet tall can be hand pulled; larger saplings can be dug or uprooted using a weed wrench or other similar tool. Plants can be repeatedly cut until no new growth appears; an immediate application of herbicide to cut stumps will prevent resprouts from forming. For mature plants, application of systemic herbicide will kill existing growth and prevent resprouts.

(top) Burning bush growth in July. Photo by Adam Gundlach, Wildlife Habitat Council. (above) Bark. Photo by James H. Miller, USDA Forest Service, Bugwood.org

(above left) Burning bush stem and leaves. Photo by Adam Gundlach, Wildlife Habitat Council. (above right) Maturing fruits with foliage beginning to turn red in fall. Photo by James H. Miller, USDA Forest Service, Bugwood.org

NOTE: Winged burning bush can be confused with native *Euonymus* species or sweetgum (*Liquidambar styraciflua*) saplings, which also exhibit corky wing-like growth on stems. Properly identify the species prior to initiating control measures.

Compiled from the U.S. Forest Service Weed of the Week, and Plant Invaders of Mid-Atlantic Natural Areas.

Privet (*Ligustrum* spp.):

Japanese privet (*Ligustrum japonicum)*
Glossy privet (*Ligustrum lucidum*)
Chinese privet (*Ligustrum sinense*)
European privet (*Ligustrum vulgare*)

Description & Biology – Privets are deciduous, perennial shrubs or small trees native to Europe, Asia, and North Africa that were introduced to North America as a landscaping hedge. There are many species of *Ligustrum* that occur throughout North America, with the ones noted above being some of the most prevalent. All of these species are similar in appearance and exhibit similar growth characteristics and life histories, making them difficult to distinguish. Depending on the species, privet grows from six to thirty feet or more in height with a spreading crown. The bark is smooth and gray or light tan in color, with green to grayish-green twigs. The leaves are dark green, opposite, ovate or elliptic, and often thick with a waxy texture. The small white flowers are borne in terminal panicles that bloom in June and July. The dark berry-like fruits mature from September through October and remain on the plant through the winter. *Ligustrum* species reproduce through seeds, root sprouts, and stump sprouts when cut. Seeds exhibit variable germination rates and are distributed widely by birds.

Chinese privet berries. Photo by James H. Miller, USDA Forest Service, Bugwood.org.

Privet species grow in a variety of habitat conditions from flood plains and forests to abandoned fields. It grows well in nutrient deficient soil given abundant sunlight, but also tolerates lower light levels in areas with richer soil. Privet is often found growing along roadsides, fence lines, forest edges, and other areas of disturbance. It can form dense, monospecific thickets that prevent growth of native vegetation.

(above left) Chinese privet foliage. Photo by Adam Gundlach, Wildlife Habitat Council. (above right) European privet growth. Photo by Nava Tabak, Invasive Plant Atlas of New England, Bugwood.org.

Control – Control measures initiated early in an invasion have the greatest potential for success. Well-established stands of privet are difficult to completely eradicate. Seedlings and young plants can be hand pulled or dug. Root fragments left in the soil may sprout to form new plants. Repeated mowing or cutting can be employed in environmentally sensitive areas where herbicide is not a viable option. Cutting or mowing should be performed as close to ground level as possible and repeated as necessary to control new growth, at least once or more per growing season.

Herbicide application is the most effective method for controlling privet infestations. Foliar sprays of glyphosate, triclopyr, or metsulfuron can be applied to dense infestations where damage to non-target species is not a concern. Cut-stump or basal bark applications of glyphosate or triclopyr can be made in areas where individual plants are scattered or desirable vegetation is growing in close association with privet.

(above left) Glossy privet leaves and flowers. (middle) Glossy privet bark. (above right) Japanese privet leaves and flowers. Photos by James H. Miller, USDA Forest Service, Bugwood.org.

European privet flowers. Photo by Nava Tabak, Invasive Plant Atlas of New England, Bugwood.org.

Compiled from the Southeast Exotic Pest Plant Council Plant Manual, and The Nature Conservancy Element Stewardship Abstract.

Bush honeysuckle (*Lonicera* spp.):
Fragrant (or winter) honeysuckle (*Lonicera fragrantissima*)
Amur honeysuckle (*Lonicera maackii*)
Morrow's honeysuckle (*Lonicera morrowii*)
Standish's honeysuckle (*Lonicera standishii*)
Tatarian honeysuckle (*Lonicera tatarica*)
Bell's (or pretty) honeysuckle (*Lonicera* x *bella*)
European fly-honeysuckle (*Lonicera xylosteum*)

(above left) Amur honeysuckle bush. Photo by Adam Gundlach, Wildlife Habitat Council. (above right) Tatarian honeysuckle bush with red flowers. Photo by Patrick Breen, Oregon State University, Bugwood.org.

Description & Biology – Non-native bush honeysuckle species are deciduous, multi-branched, upright shrubs ranging in height from six to 15 feet or more in height. Leaves are borne in opposite pairs on short stalks and are one to three inches long. Leaves generally emerge early in the spring prior to native species and remain on plants later into the fall. Pairs of tubular flowers are arranged in leaf axils along the stems. Flowers are fragrant and range in color from white and yellow to pink, and even crimson in certain tatarian honeysuckle varieties. While in bloom, non-native honeysuckle species can be distinguished from native species by the hairy flower styles – tube portion of female flower parts. Non-native bush honeysuckles produce a large quantity of fruit, which is readily eaten by many birds, dispersing seed to new locations. The fleshy, berrylike fruits range in color from bright red to orange and occasionally yellow, and contain many seeds. Native *Lonicera* species typically have blue or black berries. Bush honeysuckles reproduce through prolific seed production and vegetative sprouting where populations are established.

(top) Tatarian honeysuckle foliage and berries. Photo by Chris Evans, River to River CWMA, Bugwood.org (above) Amur honeysuckle berries in October. Photo by Adam Gundlach, Wildlife Habitat Council.

Exotic bush honeysuckles are adaptable to a variety of soil, light, and moisture conditions, and typically grow on disturbed lands. They have been observed to invade forest edges, fields, roadsides, thickets, floodplain forests, hardwood forests, grasslands, and shrublands. Although bush honeysuckles are somewhat shade intolerant, they often invade open woodlands to become a dominant component of the understory. Dense stands impede forest regeneration and decrease native plant diversity.

Control – Seedlings and young plants can be hand pulled or dug using a weed wrench (or similar tool). Larger plants can be cut repeatedly for several years until no new sprouts appear. Avoid cutting during the winter, as this encourages vigorous sprouting the following growing season. Cutting or burning dense stands will generally result in a thicker infestation due to resprouts. Dense stands of bush honeysuckle will likely require herbicide application to effectively control. Both foliar and cut-stump treatments of glyphosate or triclopyr have proven effective. The cut-stump method reduces damage to surrounding non-target vegetation and can be performed from late summer into the early winter (given appropriate conditions). Foliar applications are less time and labor intensive and are ideal for dense, monospecific stands where damage to desirable vegetation is unlikely. Foliar applications should be made late in the growing prior to fruit development. Following treatment, abundant seedlings will likely need to be controlled until the seed bank is exhausted. All control techniques will require multiple years of monitoring and maintenance due to the persistent growth of bush honeysuckles.

(above left) Amur honeysuckle foliage and flowers. Photo by Adam Gundlach, Wildlife Habitat Council.
(above right) Amur honeysuckle stem/bark. Photo by Patrick Breen, Oregon State University, Bugwood.org.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, NatureServe Explorer, and The Nature Conservancy Element Stewardship Abstract.

Chinaberry tree (*Melia azedarach*)

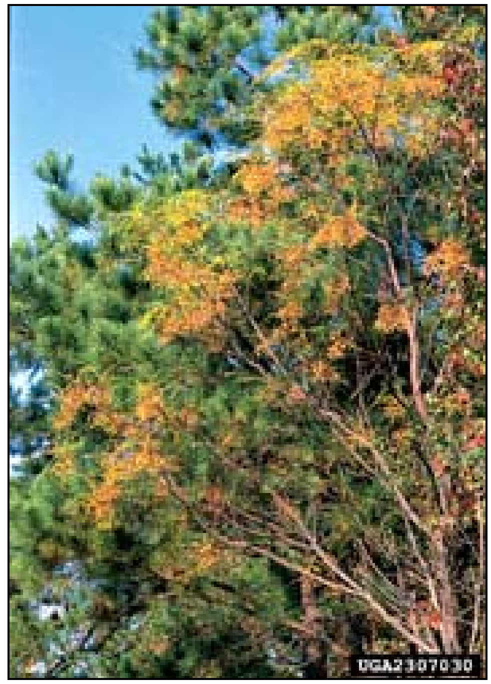
Description & Biology – Chinaberry tree, also known as umbrella tree and Persian lilac among other names, is a small to medium-sized tree in the mahogany family (Meliaceae) that was introduced as an ornamental from its native range of Southeast Asia and northern Australia. Mature trees can reach 50 feet in height and two feet in diameter with a multi-branched trunk and spreading crown. The bark is dark brown, becoming highly fissured as it matures, and the wood is soft and white. Stems are glossy green to brown, stout, and covered with many light dots called lenticels. The leaves reach two feet in length and more than one foot in width and can be bi- or tripinnately compound, comprised of many leaflets with rounded serrations around their margin. Leaflets grow up to three inches long and one inch wide, are glossy dark green above with a conspicuous light colored midvein, pale green on the underside, and turn a golden yellow in the fall. Chinaberry trees bloom from March through May, producing long clusters of fragrant, lavender to white flowers arising from leaf axils. It often initiates flowering when it reaches shrub size. Flowers develop into round single-seeded fruits, which are yellowish green when young, becoming tan to brown at maturity. Fruits are toxic to humans and livestock, but are consumed and dispersed by birds. It reproduces from abundant seeds and vegetatively from root sprouts.

Chinaberry flowers. Photo Chris Evans, River to River CWMA, Bugwood.org.

Chinaberry leaf form (top) and bark (above). Photos by James H. Miller, USDA Forest Service, Bugwood.org.

Chinaberry is mainly found throughout the southeast United States and has also been observed growing in the Midwest, mid Atlantic, and southwest United States. It invades disturbed habitats, typically growing along roadsides, forest edges, forest clearings, and other open areas, but has been noted to invade undisturbed habitats as well, such as forested floodplains, marshes, woodlands, and grasslands. It is tolerant of wet and dry soils and grows in partial shade, but is not tolerant of full shade. Dense thickets of Chinaberry can form from root sprouts, displacing native vegetation.

Control – Seedlings can be hand pulled. Herbicide application is likely the most effective control method for mature Chinaberry infestations, as cutting or other mechanical removal results in vigorous sprouting from the root system. Basal bark or cut-stump applications of triclopyr or glyphosate are the most viable options. Foliar spraying can also be used for smaller plants, but mature trees require a large amount of herbicide, making this impractical for dense, mature stands. All treatment areas will require continued monitoring and repeated control for several years to ensure success.



Orange, maturing fruits on a chinaberry tree. Photo by James H. Miller, USDA Forest Service, Bugwood.org.

Compiled from The Nature Conservancy Element Stewardship Abstract, U.S. Forest Service Weed of the Week, and Miller 2003.

White mulberry (*Morus alba*)

Description & Biology – White mulberry (*Morus alba*) is a tree species native to China; it was cultivated because of its value as a food source to the silk worm. As the art of silk making spread west, so did the range of the white mulberry, eventually becoming introduced into the United States in 1872. The silk industry in the United States ultimately failed; however, white mulberry did exactly the opposite, quickly becoming established and spreading through animal dispersal of its edible seeds.

A three-lobed white mulberry leaf, one of the species' varied leaf forms. Photo by Ohio State Weed Lab Archive, The Ohio State University, Bugwood.org

The tree is sometimes confused with red mulberry (*Morus rubra*) but growth conditions can usually determine which tree is which. Red mulberry is incredibly picky in terms of its habitat; it is most commonly found at the edges of forests with moist wooded slopes. White mulberry is not nearly as site-selective, other than being intolerant of shade. It most commonly is found in urban settings, along fence rows and in abandoned fields. The small tree reaches a height of 40 feet with red to purple fruit when fully mature. The leaves of mulberry trees can occur in three shapes: entire (no lobes), "mitten" (lobe on one side which causes the leaf to look like a mitten) or three-lobed. White mulberry leaves are bright green and shiny, with hairs only occurring on the prominent veins of the underside. White mulberry leaves are also smaller than red mulberry's and have round-toothed margins instead of pointed teeth.

Control – Seedlings may be pulled by hand. Larger specimens can be cut and the stumps either ground or painted immediately with glyphosate herbicide. Girdling can be effective for killing large trees and has the added benefit of creating a snag (dead standing tree), a valuable nesting and foraging resource for many native birds and other wildlife. Snags are vulnerable to blow-down; thus, girdling should only be done in areas away from human activity.

Compiled from: The US Forest Service Weed of the Week and Weeks 2003.

Princess tree (*Paulownia tomentosa*)

Description & Biology – Also known as empress tree or royal paulownia, princess tree is a medium sized, showy, deciduous tree of the figwort family (Scrophulariaceae) native to East Asia. Mature trees reach heights of 30 to 60 feet with rough, gray-brown bark that is interspersed with smooth, shiny areas. Stems and young branches are a glossy gray-brown and speckled with prominent pale lenticels. Stems are notably flattened at nodes where they connect with branches. The large, deciduous, heart-shaped (cordate) or ovate leaves area arranged in opposite pairs, growing more than a foot in length and nine inches wide on mature trees; leaves of saplings developing from root sprouts can be nearly twice as large. The leaves are green on the upper surface, pale underneath and noticeably hairy on both surfaces with hairy petioles reaching up to eight inches long. The fragrant, pale violet flowers are borne in large, upright clusters that bloom from April to May prior to the emergence of leaves. The fruit is a pointed, four-chambered, oval capsule that is one to two inches in length and a little more than half as wide. Fruits are pale green during the summer, maturing in the fall to become a brown, woody capsule that remains on the tree through the winter. As capsules dry out they split open to disseminate thousands of tiny winged seeds, which are wind- and water-dispersed. It reproduces from seed – a single tree can produce millions of seeds – and root sprouts, which may grow up to fifteen feet tall in a single season.

Princess tree is tolerant of infertile, acidic soils and drought conditions, allowing it to invade marginal habitats that often contain rare plants. It is typically found growing along roadsides, streambanks, forest edges, and other disturbed lands. Seeds germinate quickly after landing on an appropriate substrate and seedlings develop quickly, reaching the flowering stage within ten years.

Princess tree branch with mature (brown) and developing fruits (green). Photo by Adam Gundlach, Wildlife Habitat Council.

(top) Princess tree. (above) Close up of leaves and developing fruits in July. Photos by Adam Gundlach, Wildlife Habitat Council.

Control – Princess tree resembles native *Catalpa* species, which have similar leaves and flowers, but produce long slender bean pods. Various methods can be used to control princess tree. Young seedlings can be pulled by hand after reaching a large enough size to grasp. Root fragments remaining in the soil may sprout to form new plants. Trees can be cut at ground level after they have begun to bloom, or larger trees can be girdled to kill above ground growth. Cutting and girdling will result in numerous root sprouts and will necessitate repeated cutting or an herbicide application to new sprouts. Foliar applications of systemic herbicide, such as glyphosate or triclopyr, can be made to sprouts, saplings, and small trees in areas where damage to non-target species is not a concern. More targeted herbicide applications can be made via cut-stump, basal bark, or hack-and-squirt methods.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, and Southeast Exotic Pest Plant Council Plant Manual.

Amur corktree (*Phellodendron amurense*)

Description & Biology – Amur corktree is a deciduous tree of the citrus family (Rutaceae) introduced from East Asia as an ornamental species. Mature trees reach heights of 35 to 50 feet with spreading crowns that can be nearly as wide. Its name derives from the thick, corky bark that exhibits a slightly spongy feel. Bark on young trees is light golden brown, becoming grayish brown on furrowed on mature trees. Its inner bark has a characteristic bright yellow layer. The leaves are oppositely arranged and pinnately compound with five to thirteen dark green, slender leaflets, which turn bright yellow in the fall. Crushed leaves have a skunky citrus odor. Male and female flowers are borne on separate trees (dioecious), which reach reproductive maturity at three to five years of age. Both produce similar hanging clusters of yellow-green to maroon flowers that bloom from May through June. Female trees produce abundant clusters of fruits from mid-June through July. The small berry-like fruits, each containing five seeds, begin bright green and become black towards the end of the summer, persisting on trees into the early winter. Many birds feed on the fruits and disperse the seeds to new locations. Reproduction is mainly through seed, though trees will resprout from the roots following disturbances such as cutting.

(top) Amur corktree foliage and fruits. Photo by Patrick Breen, Oregon State University, Bugwood.org (above) Close up of bark. Photo by Chris Evans, River to River CWMA, Bugwood.org.

Amur corktree adapts to a variety of environmental conditions and hardiness zones. It grows well in full sun with moist, well-drained soil, but tolerates many soil-moisture-light combinations, including full shade. It has escaped urban plantings in the mid-Atlantic and New England to invade oak and hickory forests, suppressing regeneration of these native species. Its ability to grow under a closed canopy allows it to outcompete seedlings of native plants. The fruit of corktree provides less energy and nutritional value than tree species and does not persist through the winter like the mast of these species. Invasion by corktree can have significant impacts on wildlife populations that depend on mast-producing trees. It often is found growing in forests and riparian areas on the periphery of urban centers.

Mature Amur corktree. Photo by Richard Webb, Garden Restoration, Bugwood.org..

Control – Seedlings can be pulled by hand. Once established, corktree will require multiple years of treatment and monitoring, as seeds remain viable in the soil for several years. Removing all female trees will control the spread of this species, as male trees do not produce seed. The most effective method for controlling mature trees is through cutting or girdling followed by an immediate application of systemic herbicide (e.g. glyphosate, triclopyr), or the hack-and-squirt method, which doesn't require the tree to be completely girdled. Cutting alone will need to be repeated numerous times until no sprouts develop from the root system.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and U.S. Forest Service Weed of the Week.

White poplar (*Populus alba*)

Description & Biology – White poplar, also called silver-leaved (silverleaf) poplar, is a large, deciduous tree of the willow family (Salicaceae) that is native from central and southern Europe to central Asia. Mature trees grow to heights of 70 feet or more and two feet in diameter. Younger trees have smooth, greenish-white bark that becomes darker and rough with maturity. Young stems and twigs are green to brown in coloration and covered in dense, woolly hair, especially towards their tip. The leaves vary from oval to maple-leaf in shape with three to five broad lobes or teeth. They are dark green on top and the underside is covered in dense white hairs. Male and female flowers develop in catkins on separate trees (dioecious) that bloom between March and April. The flowers produce thousands of small seeds in late spring that are surrounded by a cottony fiber, which allows the seeds to be dispersed long distances by wind. Its seeds exhibit a low germination rate, but once established it spreads by abundant root suckers that develop from adventitious buds on lateral roots, which can quickly form a dense colony.

White poplar prefers locations that receive abundant sunlight, such as forest edges, fields, and wetland perimeters. In these locations, it can form dense colonies that interfere with natural community succession by reducing the sunlight, nutrients, water, and space available to native plants.

Control – Seedlings and young plants can be hand pulled or dug, removing as much of the root system as possible to avoid resprouts from root fragments. Mature trees can be removed by mechanical means, such as cutting, but white poplar resprouts vigorously following damage and will require repeated control of new sprouts. Prescribed burns can be effective in certain situations, but will need to be repeated or combined with cutting to achieve complete control. Systemic herbicide (e.g. glyphosate, triclopyr) can be applied via many different techniques, such as foliar spray, cut-stump, hack-and-squirt, or basal bark application, to efficiently control white poplar and prevent resprouts.

(top) White poplar leaves and stems. (above) Catkins. Photos by Paul Wray, Iowa State University, Bugwood.org.

Compiled from the PCA Alien Plant Working Group Fact Sheet, and U.S. Forest Service Weed of the Week.

Bradford (Callery) pear (*Pyrus calleryana*)

Description & Biology – Bradford (callery) pear is a deciduous tree introduced from Asia. It is widely used in landscaping. The name 'Bradford' comes from the name of the first commercial cultivar of *Pyrus calleryana*; it was marketed as a sterile tree. Subsequent introductions of other cultivars led to cross-pollination and successful reproduction. The tree now outcompetes native plants, producing dense stands along forest edges and roadsides.

Prolific clusters of small (1/2-1 inch) white flowers make this tree stand out in April and May, especially along roadsides and in suburban landscaped developments. In fall the trees are also highly visible, with red foliage and clusters of brown fruits. Other identifying characteristics are a teardrop-shaped growth pattern (pictured top right) and glossy dark green leaves with wavy, slightly-toothed margins (pictured bottom right). Leaves are alternately arranged. Bark is cherry-like and develops shallow furrows with age. Bradford pears grow quickly, reaching heights of about 40 feet. They are susceptible to breaking and splitting.

(top) Typical teardrop shape and white blooms
(bottom) Wavy leaf margins and flowers
Top by Dan Tenaglia, Missouriplants.com, Bugwood.org;
Bottom by James H. Miller, USDA Forest Service, Bugwood.org

Control – Bradford pears vigorously resprout from stumps and roots when cut (sometimes even when the original cut is painted with herbicide). Cut trees must thus be monitored carefully; resprouts must be cut or treated with herbicide until energy reserves are exhausted. Triclopyr or glyphosate can be used according to label directions in cut stump, hack-and-squirt or basal bark herbicide applications. Public education is also a critical component for control of this species, as it is still widely planted.

Compiled from the Center for Invasive Species and Ecosystem Health and the Maryland Invasive Species Council.

Common buckthorn (*Rhamnus cathartica*)

Typical orange coloration of common buckthorn underbark. Photo by Chris Evans, River to River CWMA, Bugwood.org

Description & Biology – Common buckthorn is a deciduous shrub or small tree native to Euarasia that grows from six to more than 20 feet in height with trunks reaching close to one foot in diameter. Mature plants have an irregular spreading crown. The alternate leaves are elliptic to ovate, smooth, dull green, finely serrated along the margin, and occasionally pointed at the tip. Three to four pairs of veins extend the length of each leaf. Leaves remain on plants late into the fall after most other deciduous plants have dropped their leaves. The bark varies from gray to dark brown or black and is rough with prominent lenticels. Twigs are smooth and often tipped with a sharp spine. Common buckthorn is dioecious, with male and female flowers forming on separate plants. The fragrant clusters of two to six yellow-green flowers bloom from May through June, developing in leaf axils. Abundant round, black, berry-like fruits (drupes) ripen from August through September and are consumed by a variety of birds and small mammals, which then disperse the seeds widely. Three to four grooved seeds are contained in each fruit, which often persists on the plant into the winter. Though most reproduction is achieved through seed production, buckthorn will resprout vigorously from its roots following removal of above ground growth. Glossy buckthorn (*Rhamnus frangula*), a close relative of common buckthorn and an equally problematic invader, can be distinguished by its lack of a spine at the tip of twigs, untoothed leaf margins, and hairy leaf undersides.

Buckthorn prefers full sun to moderate shade conditions found in open woodlands, canopy gaps, forest edges, fields, and prairies. It has a long growing season compared with native plants, exhibits rapid growth, and can quickly form dense, even-aged thickets that displace native vegetation. Dense stands of buckthorn seedlings, which often develop under female plants, prevent regeneration of native species. In fire-adapted communities, such as prairies and savannas, invasion by buckthorn reduces the amount of vegetation growing beneath it and subsequently alters fire cycles due to insufficient fuel loads.

(above left) Common buckthorn stem and leaves. (above right) Buckthorn berries. Photos by Paul Wray, Iowa State University, Bugwood.org.

Control – A variety of methods are available for control of buckthorn. Seedlings and saplings can be hand pulled or dug with a weed wrench. Soil disturbance should be limited during removal to prevent buckthorn seeds in the soil from germinating. Prescribed burns have proven effective at reducing and eventually eliminating buckthorn infestations from fire-adapted communities. Burns should be conducted from late March to early May and repeated in successive years as necessary to eliminate resprouts from mature plants and the seed bank. If sources of new seed exist near the treatment area, burned ground may actually increase the likelihood of animal-dispersed seeds establishing in the treatment area. Cut-stump, hack-and squirt, and basal bark herbicide treatments have all shown good results at controlling buckthorn.

Compiled from the PCA Alien Plant Working Group Fact Sheet, The Nature Conservancy Element Stewardship Abstract, and U.S. Forest Service Weed of the Week.

Black locust (*Robinia pseudoacacia*)

Black locust flowers and leaves. Photo by Bill Cook, Michigan State University, Bugwood.org.

Description & Biology – Black locust is a large, fast growing, nitrogen-fixing tree that is native to the United States, but can exhibit invasive tendencies. Two discontinuous regions form its native range; the eastern region extends along the Appalachian Mountains from Pennsylvania to northern Alabama and Georgia, while the western region stretches from southern Indiana and Illinois into the Ozark region of Missouri and Arkansas. It has been widely planted outside of its original range and is now naturalized throughout much of the country.

Black locust reaches a height of 40 to 100 feet at maturity with furrowed, dark brown bark that is characterized by deep, flat-topped ridges; saplings have smooth, green bark. Young trees develop large, paired thorns. The compound leaves alternate along stems and are composed of up to 21 leaflets, which are ovate or rounded, thin, dark green above and pale underneath. The fragrant white flowers bloom from May to June after leaf emergence, and the top petal of each flower has a yellow spot. Flowers are borne in large, showy, drooping clusters that grow from the leaf axils. The fruit is a brown, flattened seedpod that is two to four inches long and contains four to eight seeds. Seedpods ripen in September and October and remain on the tree where they open to disperse seeds from September until the following spring. Despite abundant seed production, few seedlings develop under natural conditions due to a tough, impermeable seed coat that requires scarification to initiate germination. As a result, most natural reproduction is accomplished by root sprouts, which develop from adventitious buds on the root system and at the base of the trunk.

Black locust is an early successional species that grows best in open, well-drained locations receiving full sun, as it is shade intolerant and does not compete well with other vegetation. It is often observed growing in old fields, degraded woodlands, forest edges, and roadsides. Once introduced into an area, it can quickly spread through root suckering to form dense cloned thickets connected by the root system. The shade created by thickets allows little ground vegetation to grow beneath. In the case of prairie or savanna habitat that has been

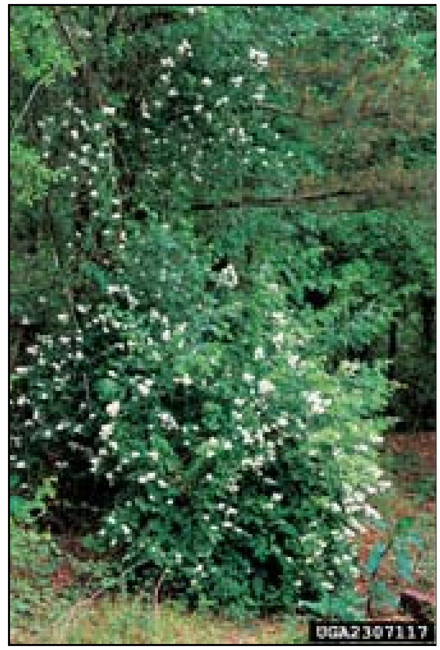
(above left) Bark of a mature black locust tree. (above right) Seedpods and leaflets. Photos by Paul Wray, Iowa State University, Bugwood.org.

invaded by black locust, the lack of ground vegetation alters the natural fire disturbance regime, which is integral to maintaining fire-adapted communities. Black locust tolerates a variety of soil conditions, but does not grow well on poorly drained, excessively dry, or compacted soils. It has been planted for erosion control and mine reclamation projects due to its dense network of roots and nitrogen-fixing ability.

Control – Cutting, mowing, or burning alone will not eradicate black locust, but may limit the spread of new shoots from the parent plant (colony). Application of systemic herbicide is the only effective method for controlling an established colony. A variety of herbicides and application methods can be employed with varying success, including foliar spray, basal bark, cut-stump, or hack-and-squirt. Repeated treatments will likely be needed in ensuing years to completely kill the root system and eradicate a colony. Annual monitoring will be needed because even colonies that appear dead have been observed to sprout again several years following herbicide application. Black locust also is susceptible to insect damage by the locust borer (*Megacyllene robiniae*) and the locust leafminer (*Odontota dorsalis*), among other insects. Larvae of the locust borer feed by burrowing tunnels through the wood, which weaken the wood and allow access points for heart fungi that cause decay. Outbreaks of the leaf miner can result in complete defoliation of black locusts in a region, and may result in mortality.

Compiled from the PCA Alien Plant Working Group Fact Sheet, The Nature Conservancy Element Stewardship Abstract, and U.S. Forest Service Fire Effects Information System.

Multiflora rose (*Rosa multiflora*)



(top) Multiflora rose creeping into canopy of a tree. (above) Young stems on the left compared to a mature stem on the right. Photo by James H. Miller, USDA Forest Service, Bugwood.org

Description & Biology – Multiflora rose, also called rambler rose, is a perennial, thorny shrub of the rose family (Rosaceae) native to East Asia that was introduced in the late 1800's as an ornamental species. It has since been planted for erosion control, as a "living fence" to control livestock, for its perceived wildlife value, and as a visual barrier along highways. It grows to 15 feet in height and nearly as wide. The stems, called canes, often climb to greater heights in surrounding vegetation via a trailing growth habit. The initial five or six feet of stems arising from the root collar are erect and then arch back towards the ground. The smooth, green stems are covered by sturdy, recurved thorns that make the plant unpleasant to manage by hand. Pinnately-compound leaves are comprised of five to eleven leaflets with sharp, finely serrated margins. Fragrant white to pinkish flowers bloom form May through July. The flowers may arise singly or in clusters and are accented in their center by bright yellow anthers. Small, red (initially yellow-green), spherical fruits, called rose hips, develop in mid- to late-summer at the end of short stalks. The rose hips become tough as they mature, persisting on the plant through the winter. The fruits are readily eaten by birds, which widely disperse the seeds and increase the likelihood of germination. A single multiflora rose plant can produce an estimated 500,000 to 1,000,000 seeds per year that may remain viable for up to 20 years in the soil. In addition to seed, it also reproduces vegetatively; the tips of arching stems can develop roots when contacting the ground to form new plants (called layering), and new sprouts can develop from the root system.

Multiflora rose grows well in a variety of environmental conditions and exhibits prolific growth after becoming established. It tolerates both full sun and shade, various soil and moisture combinations, except for standing water, and can be observed invading habitats ranging from dense forests and streambanks to prairies, fields, and roadsides. Young plants grow slowly during the first two years of development, but quickly expand thereafter to form dense, impenetrable thickets that prevent growth of native vegetation and significantly decrease habitat value for wildlife.

Multiflora rose flowers and leaves. Photo by James H. Miller, USDA Forest Service, Bugwood.org.

Control – For small initial populations or scattered individual plants, repeated cutting or mowing will prevent seed production and control the spread of the plant. Cutting should be performed a minimum of once per growing season (multiple times is preferable) as close to ground level as possible. The long arching stems and abundant thorns make cutting large plants a difficult endeavor. Established populations of multiflora rose are difficult to control due to its long-lived seedbank and vigorous resprouting following disturbance. Herbicide treatments are the most effective method to attain control. Systemic herbicides such as glyphosate and triclopyr can be applied via foliar spray, cut-stump, or basal bark methods. Treatment areas will require repeated monitoring for many years to ensure complete control, as seeds may remain dormant for up to 20 years in the soil.

Compiled from the PCA Alien Plant Working Group Fact Sheet, The Nature Conservancy Element Stewardship Abstract, and Southeast Exotic Pest Plant Council Plant Manual.

Wineberry (*Rubus phoenicolasius*)

Description & Biology – Wineberry, also called wine raspberry or Japanese wineberry, is a perennial shrub in the rose family (Rosaceae) native to China and Japan that is similar in appearance to native blackberry and raspberry species. The long, upright, arching, stems (canes) can reach nine feet or more at maturity and are covered in small spines and long, shaggy red hairs that give canes a reddish appearance from a distance. Each hair has a small spherical gland on its tip, visible if the cane is held up to the light or under a magnifying glass. Wineberry's leaves are comprised of three serrated, heart-shaped leaflets that are green above with purple veins and covered in silvery white hairs underneath. It blooms in late spring and early summer, producing small greenish flowers with white petals and reddish hairs. The fruit is raspberry-like – similar to other *Rubus* species – becoming bright red as they ripen in mid-summer. Fruits are edible and are eaten by many birds and other animals (including humans), which disperse the seeds to new locations. Wineberry reproduces by seed, vegetative sprouting from root buds, and by layering – forming new plants where cane tips contact the soil.

(top) Wineberry foliage and hairy stems. (above) Wineberry patch. Photos by Adam Gundlach, Wildlife Habitat Council.

Wineberry tends to grow in moist soils in full or partial shade. It often invades forests, fields, riparian habitats, wetland edges, savannas, and prairies, where its vigorous growth forms dense thickets over large areas that displace native vegetation. Though birds and small mammals may use wineberry thickets for nesting and cover habitat, the benefits are outweighed by the overall decrease in biodiversity associated with dense infestations.

Control – Small patches of wineberry can be removed manually by hand pulling or using a spading fork when soil is moist. It can also be cut, mowed, or burned for several consecutive seasons until no new growth appears. Cutting and mowing should be performed multiple times per growing to reduce vigor of the plant. Application of systemic herbicide, such as glyphosate or triclopyr, is effective for large infestations. Dense thickets lend themselves to foliar application, but wineberry can also be treated with cut-stump applications in the fall.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week and the Mistaken Identity guide published by the Delaware Department of Agriculture.

Japanese spiraea (*Spiraea japonica*)

Description & Biology – Also called Japanese meadowsweet, Japanese spiraea is one of numerous *Spiraea* species in the rose family (Rosaceae) that have been cultivated for use in gardening and landscaping for their showy pink flowers. It is a small, deciduous, perennial shrub that grows four to six feet in height and similar in width. The erect, round, slim stems are brown or reddish-brown in color and are sometimes hairy. The alternate leaves are one to three inches long, egg-shaped tapering to a point, and have toothed margins. The flowers bloom from June through July, arising from the tip of branches in showy pink clusters. Seeds are produced in small, smooth capsules from July through August.

Japanese spiraea tolerates a variety of soil conditions, grows in partial shade to full sun, and often colonizes disturbed areas. It is commonly found growing along stream and river margins, as its seeds are often dispersed by water, and it also inhabits forest edges, successional fields, roadsides, and rights-of-way. Once established, it can spread to form thickets in surrounding meadows and forest gaps. The dense growth crowds out native herbaceous and shrub species.

(top) Japanese spirea shrub. Photo by Adam Gundlach, Wildlife Habitat Council. (above) Leaves and flowers. Photo courtesy of Great Smoky Mountains National Park Resource Management Archives, USDI National Park Service, Bugwood.org.

Control – Repeated cutting or mowing of shrubs is recommended for small patches or locations where herbicide is not an option. Cutting or mowing should be performed at least once per growing season prior to seed production. Cuts should be made as close to ground level as possible. This method will need to be repeated for several years until energy reserves are exhausted, as spiraea resprouts from the root collar following cutting.

Foliar herbicide applications of glyphosate or triclopyr can be used for dense thickets of Japanese spiraea where damage to non-target species is not a concern. More targeted applications can be made via the cut-stump method. Applications can be made year-round given temperatures of 65 degrees Fahrenheit or above to ensure adequate absorption of herbicide by the plants.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, and Southeast Exotic Pest Plant Council Plant Manual.

Siberian elm (*Ulmus pumila*)

Description & Biology – Siberian elm, also called Chinese elm, dwarf elm, and Asiatic elm, is a fast-growing deciduous tree native to China, Korea, and eastern Siberia. This hardy member of the elm family (Ulmaceae) reaches a height of 50 to 70 feet at maturity with a rounded, spreading crown and rough gray to brown bark that shallow, irregular fissures with age. The alternate leaves are between one and three inches long, dark green above, paler underneath, and pointed at the tip. Leaf margins are serrated with small teeth and unlike other elm species, the base of Siberian elm leaves are nearly symmetric, joining together in a "V". Small, drooping clusters of green flowers bloom in March and April, sometimes appearing before leaves begin to develop. The fruit is a small, circular, flattened samara containing a single seed in the center. It reproduces solely by seed, which is wind-dispersed.

Siberian elm tree. Photo by Patrick Breen, Oregon State University, Bugwood.org.

Siberian elm tolerates a range of environmental conditions and is resistant to Dutch elm disease. It grows best in full sun in fertile well-drained soils, but will also grow in moist soils typical of riparian areas as well as arid locations. It often invades dry to mesic prairies and stream banks. Once a seed-producing tree is established, the fast-growing seedlings form thickets under the parent plant, which crowd out native vegetation and increase the likelihood of invasion by other weed species. Siberian elm produces seed early in the spring and seeds exhibit a high germination rate that gives the species a competitive advantage over other vegetation.

Control – Successful control of Siberian elm requires prevention of seed production. Mature seed-producing trees can be cut down or girdled. Trees that are cut down will sprout from the stump. The sprouts can be repeatedly cut or treated with a cut-stump application of herbicide, such as glyphosate or triclopyr. Cut-stump treatments should be performed during the summer to avoid the spring sap flow, which prevents adequate transport of herbicide to the roots. Basal bark treatments of triclopyr can also be used for control. Girdling, when performed correctly, will kill the tree within one to two growing

(above left) Siberian elm stem and leaves. Photo by Steve Dewey, Utah State University, Bugwood.org. (above right) Samaras containing seeds. Photos by USDA NRCS Archives, USDA NRCS, Bugwood.org.

seasons and prevent resprouts from forming. A three- to four-inch wide band of the outer bark should be removed by making parallel cuts around the circumference of the trunk. Then use a blunt object to peel away the bark between the cuts. Avoid damaging the inner wood (xylem) because this will trigger resprouting. Seedlings can be pulled by hand and saplings can be dug or removed using a weed wrench. Prescribed fire can be employed in fire-adapted communities to kill Siberian elm saplings.

Compiled from the PCA Alien Plant Working Group Fact Sheet, U.S. Forest Service Weed of the Week, and NRCS Plant Guide.

AQUATIC INVASIVE SPECIES

Adapted from PCA Alien Plant Working Group – Mid-Atlantic List

Alligatorweed (*Alternanthera philoxeroides*)

Description & Biology – Native to South America, alligatorweed is an aggressive, perennial pest of waterways from coastal Virginia to Florida and west to Texas, with a disjunct population in California. It exhibits both aquatic and terrestrial growth and roots in wet soils or shallow water along streams, drainages, and other waterways, forming dense mats of vegetation that extend from the shore out over open water. The stems are hollow (except at the nodes) when growing aquatically, but often contain solid pith when growing terrestrially. The opposite leaves are sessile, ovate to lanceolate, and have a prominent midrib. Small, white, clover-like flowers bloom from April through October and develop on short stalks arising from leaf axils near the end of stems. Alligatorweed produces few seeds and most are sterile, though viable seeds have been found in the United States. Reproduction is achieved vegetatively through stem fragments and rhizomes. Roots are produced at each node along stems, and if stem fragments are separated from the main plant, they can easily be transported by water to new locations, where they can become established as new plants.

Alligatorweed flower. Photo by Gary Buckingham, USDA Agricultural Research Service, Bugwood.org.

Alligatorweed displaces native plants in ditches and along banks of waterways and shades out submerged aquatic vegetation, causing decreased oxygen levels in water below mats of vegetation. The dense mats increase sedimentation and impede water flow in drainages, which may lead to flooding during storm events. They can also impede navigation of small waterways or access to shoreline in larger water bodies.

(top) Alligatorweed flower and leaves. (above) Waterway being choked off by alligatorweed. Photos by Chris Evans, River to River CWMA, Bugwood.org.

Control – Several biological control agents have been identified and released in the United States to control alligatorweed, including the alligatorweed flea beetle (*Agasicles hygrophila*), alligatorweed thrip (*Amynothrips andersoni*), and the alligatorweed stem borer (*Arcola malloi*). They have proven effective in reducing or eliminating alligatorweed infestations in the southern United States. Unfortunately, the insects exhibit poor survival in northern portions of the alligatorweed's range, such as Virginia, and must be imported each season to maintain populations sufficient for control.

Herbicides labeled for use in aquatic habitats, such as Rodeo®, may be used to control alligatorweed. The herbicide will only be effective when applied to emergent vegetation. Oxygen depletion may result following treatment, as the dead plant matter decomposes.

Compiled from Van Driesche et al. 2002, and North Carolina State University Aquatic Weed Fact Sheet.

Brazilian waterweed (*Egeria densa*)

Description & Biology – Brazilian waterweed, also called common waterweed, South American waterweed, and Brazilian elodea, is a perennial, submergent freshwater weed common in the aquarium trade that has been introduced to water bodies throughout much of the United States. It typically roots in the muddy bottom substrate of water bodies, but can occur as free-floating plants when dislodged. The slender, cylindrical stems usually grow from one to two feet in length, but can reach up to 15 feet or more. The bright green leaves are arranged around the stem in whorls of three to six leaves, with the lowermost leaves in opposite pairs. The flowers, if present, are composed of three white petals held just above the water that bloom from summer through fall. It rarely is observed to produce seed, reproducing instead vegetatively through stem fragments. Double nodes, consisting of two closely spaced single nodes, located every six to twelve inches along the stem produce lateral buds, branches, and adventitious roots. Any stem fragment containing a double node can root to form a new plant.

Brazilian waterweed grows in lakes, ponds, ditches, and slow-flowing streams and rivers. It forms dense, monospecific mats that cover large areas, preventing growth of native aquatic vegetation and decreasing overall biodiversity of aquatic environments.

Control – Brazilian waterweed may be mistaken for native waterweed species (*Elodea* spp.) or hydrilla (*Hydrilla verticillata*). It can be distinguished by its finely-toothed leaf margins (visible with a 10x magnifying lens), compared to the conspicuously-toothed leaf margins of hydrilla and the smooth leaf margins of native *Elodea* species. Brazilian waterweed generally has longer leaves, up to four centimeters, than hydrilla or *Elodea* species, which typically grow to less than two centimeters. *Eldoea* species can also be distinguished by its whorls of three leaves compared with the four to six leaves per whorl typical of Brazilian waterweed and hydrilla. Brazilian waterweed flowers are also larger, growing to more than 1.5 centimeters in diameter, whereas hydrilla and *Elodea* flowers remain below one centimeter in diameter.

(top) Brazilian waterweed plants. Photo courtesy of Virginia Tech Weed Identification Guide Archives, Virginia Polytechnic Institute and State University, Bugwood.org. (above) Close up of stem and leaves. Photo by Richard Old, XID Services, Inc., Bugwood.org

Mechanical methods of removal, such as cutting, pulling and digging, will likely only serve to spread plant fragments and increase an infestation.

Covering small, localized patches with opaque fabric sheeting to prevent sunlight from reaching the plants is an option. For water bodies where the inflow and outflow can be monitored and controlled, sterile grass carp may be introduced to reduce Brazilian waterweed populations, as grass carp find the plant highly palatable. A drawback of this method is that grass carp do not selectively focus on waterweed and will eat other desirable vegetation as well. Certain aquatic herbicides have also been used to control waterweed, such as Diquat dibromide, endothal, and fluridone (Sonar®). Before employing chemical control measures, be sure to read all labeling thoroughly, check local and state restrictions on herbicide use in aquatic environments, and consider the impact such applications will have on other aquatic vegetation and organisms.

Compiled from the Invasive Plant Atlas of New England, University of California-Davis database, and Washington State Department of Ecology.

Common water hyacinth (*Eichhornia crassipes*)

Description & Biology – Water hyacinth is a free-floating aquatic plant that has invaded many aquatic environments in the eastern and southern United States since its introduction from South America. Its thick, waxy, glossy leaves are four to eight inches in diameter, elliptic to circular, often with an inward curve or undulation, and contain a dense network of longitudinal veins. In open conditions, whorls of six to ten leaves form a bulbous basal rosette resting just above the water surface, but in crowded conditions, the leaves are held high above the water on spongy, upright stalks that can reach more than two feet in length with a bulbous base. Leaves and roots are borne at nodes along the rhizome, which can be nearly three inches in diameter and up to 12 inches long. The adventitious roots can vary in color from dark purple or bluish to light pink-violet. The fibrous root system can comprise half of the plant's biomass. Showy, lavender flowers are borne on a flower stalk in a terminal cluster (spike) of four to 25 flowers. At the end of the flowering cycle, the flower stalk bends over to submerge the flowers and release the fruit capsules, which contain up to 450 seeds. Although, water hyacinth produces abundant flowers, there have been few observations of seed production or seedlings. The main form of reproduction is vegetative. The dense mats typical of hyacinth infestations are formed by production of daughter plants from stolons.

(top) Water hyacinth flowers and leaves. Photo by Wilfredo Robles, Mississippi State University, Bugwood.org. (middle) Flowering plant. Photo by John D. Byrd, Mississippi State University, Bugwood.org. (above)

Water hyacinth exhibits aggressive growth in a variety of habitats, including ponds, lakes, rivers, marshes, and wetlands. It grows best in warm, neutral waters with high nutrient content, but tolerates fluctuations in water level, nutrient level, flow, and acidity. It can rapidly form dense mats that prevent sunlight from reaching submerged vegetation. The resultant decline in aquatic plant diversity has a cascading effect through the food chain and results in an overall decrease in biodiversity. Hyacinth infestations clog waterways and interfere with navigation, recreation, irrigation, and drainage, which can result in problems with flooding during storm events.

Control – Small, initial populations of water hyacinth may be controlled by manual removal. All plant parts must be removed to prevent new infestations from forming, as any plant fragment may develop into a new plant. Herbicide applications are the most effective method for controlling large expanses of hyacinth. Glyphosate (Rodeo®) and 2,4-D have both been used to control hyacinth, though 2,4-D has a greater toxicity to other aquatic organisms than does glyphosate. Both herbicides are non-selective and will damage any plants they contact, including desirable native species. Herbicide applications may also be coupled with biological control agents to increase efficacy. Three such agents were released in southern states in the 1970s, including two water hyacinth weevils – *Neochetina bruchi* and *Neochetina eichhorniae* – and the water hyacinth moth (*Niphograpta albiguttalis)*. Several other biocontrol agents have since been released or are under consideration for possible introduction. *Neochetina* species have proven most effective at reducing hyacinth infestations. In contained bodies of water, sterile grass carp have also been used for control, but carp prefer other vegetation to hyacinth when available, necessitating high stocking rates. Carp should only be used when loss of native vegetation is acceptable and the carp can be contained to an area.

Compiled from The Nature Conservancy Element Stewardship Abstract, Van Driesche et al. 2002, and Southeast Exotic Pest Plant Council Plant Manual.

Hydrilla (*Hydrilla verticillata*)

Hydrilla. Photo by Raghavan Charudattan, University of Florida, Bugwood.org.

Description & Biology – Hydrilla is a perennial, submerged aquatic plant native to Asia and possibly areas of India. It was introduced for use in aquariums and presumably escaped to natural habitats through improper disposal of aquarium material. It typically roots in bottom substrate, though fragments or entire plants can survive in a free-floating state, and exhibits varied growth forms depending on the environmental conditions and water depth. In deep water, stems may extend to more than 30 feet in length. The leafy stems rarely branch until reaching the water surface, where they produce numerous horizontal branches. The small, lanceolate leaves (3/4-inch long) have finely toothed margins, red veins, and sometimes fine teeth along the midrib. Leaves are arranged in whorls of four to eight that give the appearance of a bottlebrush. Hydrilla may be monoecious (male and female flowers produced on same plant) or dioecious (male and female flowers produced on separate plants). The monoecious form is typically found in the mid-Atlantic United States, while the dioecious form is typically found in the southeastern United States as well as California and Texas. The female flowers are composed of three white sepals and three translucent petals that arise from the tip of stems and float on the surface. Male flowers are borne on a short stalk and are composed of three sepals and three petals, both of which are whitish to reddish. At maturity, the male flowers are released to float to the surface where they release their pollen. The female flowers are then wind pollinated. Seed production and viability is low and of little importance to hydrilla reproduction. Hydrilla mainly reproduces vegetatively through stem fragments, which may sprout to form new plants, and more importantly through production of turions – specialized over-wintering buds. Turions can be produced from axils on the stem as cone-like propagules or underground on rhizomes and stolons as bulb-like "tubers." Underground turion production is considered more important, as the subterranean variety is much more prolific. Subterranean turions can remain viable in sediment for several years, out of water for several days, and may survive consumption by waterfowl or herbicide application. Turion production is highest in the fall and spring, is greater in floating plants than rooted plants, and decreases with increasing plant density.

Hydrilla infestation. Photo by Michael Frank, Galileo Group Inc., Bugwood.org.

Hydrilla grows in a variety of habitat types and water conditions including slow-flowing rivers, lakes, ponds, tidal zones, and reservoirs. It tolerates various combinations of water chemistry, temperature, sediment load, and light intensity. It forms dense monospecific stands that exclude native aquatic vegetation by dominating nutrients and sunlight, thereby reducing habitat for certain aquatic organisms. However, it has shown to improve water quality, habitat for certain fish species, and provide forage for waterfowl. The dense mats can quickly choke a body of water and impede access for boating, fishing, and other recreational uses. The stems cling to boats and, if not cleaned properly, are transported to other locations where they can form new infestations.

Control – Hydrilla can be confused with native *Elodea* species, but can be distinguished by its toothed leaf margins – *Elodea* species are generally smooth – and greater number of leaves per whorl – four to eight as opposed to three to five for *Elodea* species.

Harvesting hydrilla is only effective for small initial populations, as control is only possible if all stem fragments are collected. A single fragment containing one whorl of leaves (node) can sprout to form a new infestation. Mechanical harvesters may be used to manage expansive mats that interfere with intake pumps or other activities, but harvesting will need to be performed multiple times per growing season due to the prolific growth of hydrilla. Drawdowns have been used effectively to reduce hydrilla populations in water bodies with water control structures. Drawdowns are most effective when performed in the fall while turion development is taking place and early spring prior to initiation of new growth. Dormant turions can remain viable in the soil following drawdowns, and may lead to new infestations. Various herbicides have been used to control hydrilla, including fluridone, copper sulfate (Komeen®), endothal (Aquathol®), and bensulfuron methyl. Copper sulfate and endothal are both a non-selective, contact herbicides; copper sulfate is highly toxic to fish. Fluridone and bensulfuron methyl are both systemic herbicides that require long exposure periods, and are intended to reduce, but not necessarily eliminate hydrilla populations. For all herbicide treatments, the concentration and exposure time will determine the effectiveness. Be sure to check for restrictions on use of herbicide in aquatic habitats and thoroughly read all herbicide labeling before initiating any treatments.

Hydrilla. Photo by Chris Evans, River to River CWMA, Bugwood.org

Several biological control agents, including two fly species (*Hydrellia pakistanae* and *Hydrellia blaciunasi*) and two weevil species (*Bagous affinis* and *Bagous hydrillae*), have been released in southern states

(Florida, Georgia, Texas, and Louisiana). Both fly species established populations at their original release sites and exhibited some range expansion, though presence outside of the original release sites is minimal. Both weevil species failed to establish permanent populations. Research into new biocontrol agents is ongoing, as well as improved methods for propagating existing biocontrol agents. Sterile grass carp have been used in contained water bodies for control of hydrilla. Grass carp will feed on all aquatic vegetation, including native species, and their release may be restricted by local and state laws.

Compiled from The Nature Conservancy Element Stewardship Abstract, Van Driesche et al. 2002, Plant Invaders of Mid-Atlantic Natural Areas, and Southeast Exotic Pest Plant Council Plant Manual.

European frog-bit (*Hydrocharis morsus-ranae*)

Description & Biology – European frog-bit is an annual aquatic plant that appears to be approaching the Delaware River Basin from both the northeast and northwest. The species was introduced to North America through an arboretum in Ottawa and has since spread southward; populations currently exist from Orleans County through St. Lawrence County along New York's northwest border and in the northeast counties of Clinton, Essex and Saratoga. Given this species' pattern of southward spread since its introduction to the continent in the 1930s, managers in the Delaware River Basin should be aware that this plant could soon be a real threat. The good news is that it is not currently known to exist within the basin and can thus be prevented from becoming established if land managers are vigilant in monitoring and removing any specimens that appear.

European frog-bit with its characteristic clump of leaf stalks and three-petaled flower. Photo by Leslie J. Mehrhoff, University of Connecticut, Bugwood.org

European frog-bit looks like a miniature water lily, with heart-shaped green floating leaves and white flowers. Leaves are 1.5 to 3 inches long (in contrast to the 4- to 11-inch long leaves of the native white water lily *Nymphaea odorata*), spongy on the underside, and borne on stalks that occur in clumps. The stalks extend 1 to 3 inches beneath the water's surface. Roots extend from the stalks, forming a tangled mat that dangles in the water rather than attaching to the bed of the water body. Flowers have a yellow center with three white petals, which distinguish it from the five-petaled native little floating heart (*Nymphoides cordata*) and white water lily, which has many rows of petals. European frog-bit closely resembles American frog-bit (*Limnobium spongia*), which is native to portions of the Mid-Atlantic. The species can be distinguished by leaf characteristics: the spongy underside of American frog-bit's leaves is gelatinous and red-tinged.

European frog-bit colonizes marshes, swamps, lakes, ponds, shallow river edges, and wet ditches. It spreads vegetatively via stolons and turions (winter buds), forming dense mats that dramatically reduce light and nutrient availability for native submerged vegetation. Researchers in Canada have noted that it often occurs in wetlands that also have invasive populations of purple loosestrife (*Lythrum salicaria*).

Control – Control measures remain unknown. It is speculated that boats may be a source of spread, so careful cleaning of all equipment by boat motorists may slow dispersal. For control of established populations: judging by the species' tendency to spread via stolons and turions, it would be logical that manual removal of these parts (before the turions drop from the stolons for the winter) could be effective. Care should be taken to remove and dispose of all plants parts, as this species is known to spread via fragments.

NOTE: European frog-bit resembles American frog-bit (*Limnobium spongia*). Both species have leaves with spongy undersides, but American frog-bit leaf undersides are gelatinous and red-tinged. Another look-alike, the native white water lily (*Nymphaea odorata*), has larger leaves and complex flowers. Other native aquatics with floating leaves can be distinguished by having more than three petals, having colored flowers and/or lacking notches in the leaves.

Compiled from: Canadian Wildlife Service, U.S. Geological Survey, Maine Volunteer Lake Monitoring Program, New York Sea Grant and the USDA PLANTS Database.

Yellow flag iris (*Iris pseudacorus*)

Yellow flag iris. Photos by Adam Gundlach, Wildlife Habitat Council

Description & Biology – Yellow flag iris is a perennial herbaceous plant that was introduced to the United States about 150 years from Eurasia for ornamental purposes and soon escaped cultivation, becoming naturalized in many wetland habitats. It is still widely available, used in ornamental landscaping, erosion control, and water treatment, and can be found in varying abundance throughout most of the continental United States. The stiff sword-like leaves are smooth and green, and arise from the base of the plant, reaching more than three feet in length. Yellow flag blooms from April to June, producing multiple yellow or sometimes cream-colored flowers on a single stalk (called a peduncle). Flowers are comprised of three upward-curving petals and three larger, downward-curving sepals. The sepals often have purple or reddish-brown veins running through them. In bloom, it is easily identified by its showy yellow flowers, the only iris species in the United States with flowers of this color. Flowers give way to elliptic, angled capsules that are up to three inches long and contain more than 100 white seeds, which harden and turn brown as they mature. The seeds are dispersed by water and have been observed to exhibit a relatively high germination rate, as much as 60 percent. Yellow flag produces thick, pink, tuberous rhizomes that spread laterally to form dense clonal stands.

Yellow flag grows in various wetland habitats and saturated soils including marshes, ditches, and along the margins of lakes, ponds, streams, and rivers in up to ten inches of water. It tolerates both fresh and brackish waters and forms dense stands that exclude other native vegetation. Dense growth can raise the surrounding seedbed through increased sedimentation and a thick rhizome mat, which may alter the water table and natural succession to favor more upland species. Rhizomes are capable of surviving severe drought conditions, and may be transported to new locations during flood events, where they can form new infestations. Yellow flag offers no benefit to wildlife, as all plant parts, especially the rhizomes, contain glycosides that are poisonous to animals, and the seeds are not eaten by bird species.

Control – Young infestations have a high likelihood for control, but well established populations can be difficult to control due to its vigorous sprouting. Small populations may be managed through manually digging, cutting, or pulling, but complete control will require repeated efforts due to the plants ability to resprout from even small rhizome fragments. Disturbance caused by digging and pulling may also serve to increase an infestation by creating favorable conditions for seed germination of yellow flag and other unwanted species. Care should be taken when handling yellow flag, as the leaves and rhizomes contain a resinous substance that can cause skin irritation.

Yellow flag can be effectively controlled with herbicides labeled for use in aquatic habitats, such as certain formulations of glyphosate (Rodeo®, Aquamaster®). Glyphosate can be applied either as a foliar spray or with a dripless wick applicator or brush. Applying herbicide immediately after cutting the leaves may increase the effectiveness of the treatment and reduce effects to non-target vegetation. Late season applications are often effective for rhizomatous plants because during this time energy is being translocated to the roots and herbicide is also taken to roots, resulting in more effective kill of the plant.

NOTE: When not in bloom, yellow flag is similar in appearance to native blue flag iris (*Iris versicolor*) and cattail (*Typha*) species, which have leaves of similar form.

Compiled from The Nature Conservancy Element Stewardship Abstract, Invasive Plant Atlas of New England, and U.S Forest Service Weed of the Week.

Marsh dewflower (*Murdannia keisak*)

Marsh dewflower. Photos by Linda Lee, University of South Carolina, Bugwood.org.

Description & Biology – Marsh dewflower, also called marsh dayflower, Asian dayflower, and wartremoving herb, is a perennial herbaceous plant of the spiderwort family (Commelinaceae) native to eastern Asia that was likely introduced to the United States as a contaminant in imported rice. It has become established in coastal states from Maryland and Virginia to Louisiana, as well as in Washington and Oregon on the West Coast. Its slender, grass-like stems sprawl across the ground, rooting at nodes and reaching up to 30 inches in length. The upturned leaves alternate along stems with sheaths at their base that surround the stem. Leaves are narrow, grow one to two inches or more in length, and taper to a point at their apex. The flowers bloom from August to late Septemeber, arising from leaf axils in the upper portion of stems. Flowers are comprised of three bluish-purple to white petals. Plants are capable of producing thousands of tiny seeds, which are eaten by waterfowl and disseminated to new locations. It also reproduces vegetatively through its perennial root system, and root fragments can be dispersed by flood waters to form new populations.

Marsh dewflower grows in saturated soils along pond and stream edges, floodplains, freshwater marshes and freshwater tidal marshes. Its aggressive growth forms dense mats of vegetation that exclude native plants. The dense growth also stabilizes soil and reduces the flow of slow moving waters, which results in sedimentation and an increase in other vegetation.

Control – Small patches can be hand pulled prior to seed set. Be sure to remove all stem fragments from the site, as stem fragments left behind are capable of sprouting to form new plants; mechanical removal is not recommended because of this characteristic. Late season herbicide applications performed before seed set are effective for controlling large infestations. Use only herbicides labeled for use near aquatic habitats, such as certain glyphosate formulations (Rodeo®, Aquamaster®). Early season glyphosate treatments have proven to be ineffective at controlling marsh dewflower infestations.

Compiled from [Plant Invaders of Mid-Atlantic Natural Areas](), [Virginia Department of Conservation and Recreation Fact Sheet](), and [NatureServe Explorer]().

Parrotfeather watermilfoil (*Myriophyllum aquaticum*)

Description & Biology – Parrotfeather, also called Brazilian water milfoil, is an herbaceous aquatic plant native to the Amazon River in South America. It was introduced to the United States in the late 1800s for use in aquaria and aquatic gardens, and soon escaped cultivation, spreading through many waterways in eastern and southern states. Parrotfeather has stout stems that may reach more than 15 feet in length. The stems and submerged leaves are sometimes red in color. Tips of stems may emerge up to one foot out of the water. The fine, pinnately-divided leaves are arranged in whorls of four to six around stem nodes. Submerged leaves grow from one-half to more than an inch long, with 20 to 30 divisions per leaf. Emergent leaves can grow to two inches long and have six to 18 divisions per leaf. The inconspicuous flowers are white, arising in leaf axils, and the small fruits (if produced) are up to 1/8-inch long. Sexual reproduction is not an important means of reproduction, as only female plants are known to exist in the United States. Most reproduction is vegetative, accomplished by spread of plant fragments and new sprouts that develop from the rhizomes.

Emergent parrotfeather foliage.Photos by Alison Fox, University of Florida, Bugwood.org.

Parrotfeather grows in lakes, ponds, slow-moving streams, and drainage ditches, exhibiting aggressive growth, especially in high nutrient waters. Emergent stems can survive temporary water level fluctuations that expose them on stream banks and lake shores. Parrotfeather forms dense monospecific mats that displace and shade out native aquatic vegetation, significantly altering the aquatic food web. Its dense growth clogs waterways, interferes with irrigation, drainage, and recreational activities, and provides breeding habitat for mosquito populations.

Control – Parrotfeather may be mistaken for native coontail (*Ceratophyllum demersum*), non-native hydrilla (*Hydrilla verticillata*), or other *Myriophyllum* species, of which there are eight native species in eastern North America, as well as Eurasion watermilfoil (*Myriophyllum spicatum*) another non-native invasive species. Confirm identification prior to initiating control measures.

Due to parrotfeather's ability to reproduce from plant fragments, manual and mechanical control methods, such as cutting or rotovation (underwater tilling), may only increase an infestation by spreading fragments. Mechanical controls should only be performed on small contained water bodies, or to open channels that have already been completely clogged with parrotfeather. Water level draw downs have been used in certain situations, but results vary depending on the time of year and weather conditions, as the rhizomes are able to persist through unfavorable conditions. Herbicide may be applied to emergent parrotfeather vegetation, though complete control is unlikely. The emergent stems and leaves have a waxy outer layer that impedes herbicide uptake by the plant. Herbicides that have been used include 2,4-D. diquat, diquat with complexed copper, endothall dipotassium salt, endothall with complexed copper, and glyphosate (Rodeo®).

Parrotfeather infestation. Photo by Alison Fox, University of Florida, Bugwood.org.

Compiled from Plant Invaders of Mid-Atlantic Natural Areas, Invasive Plant Atlas of New England, Virginia Department of Conservation and Recreation Fact Sheet, and Washington State Department of Ecology.

Eurasian watermilfoil (*Myriophyllum spicatum*)

Description & Biology – Eurasian watermilfoil is an herbaceous, submerged to emergent aquatic plant native to Eurasia and northern Africa that was accidentally introduced to North America from improperly disposed aquaria contents or attached to a sea vessel. Its roots in the bottom of water bodies and the stems generally extend to the water surface, growing from three to more than 30 feet in length. The branching stems are slender, smooth, lack leaves near their base, and often form dense mats near the surface. The feathery leaves are green to grayish-green, finely divided with 12 to 16 pairs of fine leaflets, and are arranged in whorls of three to four leaves at stem nodes. Its yellow flowers bloom from July through September, forming on a spike that extends a few inches above the water. Female flowers are produced at the base of the spike and male flowers are produced at the apex of the spike. After pollination, the flower spikes often re-submerge and the female flowers form four, small, nutlike capsules, each containing four seeds. Though viable seeds are produced, sexual reproduction is not an important means of spread. Population expansion through rhizomes, stem fragmentation, and axillary buds are the main forms of reproduction. Stems nodes that contact bottom mud substrate may root to form entirely new plants.

(top) Eurasian milfoil stem and leaves. Photo by Robert H. Mohlenbrock, USDA NRCS PLANTS Database, Bugwood.org. (above) Milfoil growth in water. Photo by Alison Fox, University of Florida, Bugwood.org

Eurasian watermilfoil invades ponds, lakes, canals, and other slow moving waters in both fresh and brackish environs. It tolerates polluted waters and invades degraded or disturbed aquatic habitats where native plants do not survive, but does not spread to locations with established native plant communities. Eurasian watermilfoil generally initiates growth prior to native plants, giving it a competitive advantage. It forms dense vegetative mats near the water surface that displace or shade out native aquatic vegetation and provide breeding habitat for mosquito populations. Infestations significantly reduce overall aquatic biodiversity, including plants, invertebrates, and fish, and alter the temperature, pH, and oxygen level of the water. Thick mats also impede boat traffic and recreational activities.

Control – Several native watermilfoil species exist in the eastern United States and can easily be confused with Eurasain watermilfoil and parrotfeather. Confirm identification of the species prior to initiating control efforts.

Eurasian watermilfoil can be harvested using mechanical harvesters, but this will only temporarily reduce its prevalence and will spread stem fragments that can develop into new plants. Manual harvesting of small patches can also be performed using a rake. Harvesting should be performed multiple times per growing season to be most effective, and harvesting during peak biomass in early summer will limit subsequent growth. Where water levels can be controlled, water level manipulations can effectively reduce milfoil invasions. Drawdowns during the summer can dehydrate plants, and during the winter, given adequate cold temperatures, may result in freeze damage to the perennial roots that can kill the plant. Other control methods reduce the amount of sunlight that reaches milfoil plants through shading with native floating plants, light-limiting dyes, and shade barriers, but these techniques will also affect native plants. Milfoil has also been controlled using the herbicides fluridone (Sonar®) or 2,4-D. Fluridone is a selective herbicide for milfoil and other aquatic weeds that is available in liquid (Sonar AS®) or pellet (Sonar PR®, Sonar Q®, Sonar SRP®) forms. It is most effective when applied to stationary waterways during early stages of growth. Moving water and other factors that reduce plant-herbicide contact will limit the effectiveness. It can be applied safely in areas where swimming, fishing, and potable water intakes are present. Both liquid (DMA*4IVM®) and granular (AquaKleen®, Navigate®) formulations of 2,4-D may also be used to control watermilfoil. They are fast-acting systemic herbicides that are selective for broadleaf species, such as watermilfoil.

Milfoil infestation. Photo by Robert L. Johnson, Cornell University, Bugwood.org.

Compiled from the PCA Alien Plant Working Group Fact Sheet, Invasive Plant Atlas of New England, Van Driesche et al. 2002, and Southeast Exotic Pest Plant Council Plant Manual.

Water lettuce (*Pistia stratiotes*)

(top) Crowded waterlettuce plants. Photo by Troy Evans, Bugwood.org. (above) Mature waterlettuce with infestation in background. Photo courtesy of USDA ARS Archives, USDA Agricultural Research Service, Bugwood.org.

Description & Biology – Water lettuce is an herbaceous, floating aquatic plant that is found in tropical regions throughout the world. Its native origin is disputed, but is believed to be South America or Africa. In the United States, it mainly occurs in Gulf coast states, as it is not tolerant of cold winter temperatures, but scattered populations exist in other states, such as Virginia and New York. In warm climates it grows as a perennial, but in temperate regions it grows as an annual, regenerating from seed each spring. It grows as floating rosettes of grayish-green leaves, which occur individually or connected by stolons. The fleshy leaves have a rounded wedge shape and are covered by dense white hairs. They grow from a few inches to more than a foot in length and have conspicuous parallel veins that form ridges along their length. The inconspicuous, pale-green flowers occur in a spadix – an inflorescence of flowers contained within a large bract (called a spathe) – with several male flowers arranged on above a single female flower, separated by a constriction in the spathe. Female flowers produce green berries, which contain many small, light brown seeds. Although the seeds are viable and exhibit a high germination rate, the main form of reproduction is through vegetative shoots that develop on stolons, producing new rosettes. Multiple secondary rosettes may develop from a single parent plant to form dense, interconnected mats. Numerous feathery roots hang down from the bottom of the plant.

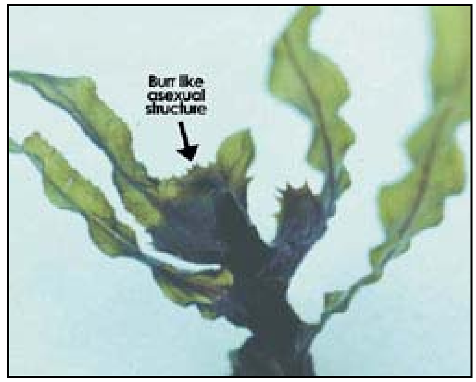
Water lettuce grows in lakes, ponds, canals and other slow moving waters. It can form extensive mats of vegetation that block sunlight from reaching submersed vegetation, altering natural biotic communities. Waterlettuce infestations may increase siltation rates, reduce oxygen levels, alter water temperatures, and add significant amounts of dead plant matter to the bottom substrate. Dense mats impede boat traffic and water flow in irrigation channels, disrupt recreational activities, and block flood control channels. Several mosquito species breed on water lettuce and are potential disease vectors.

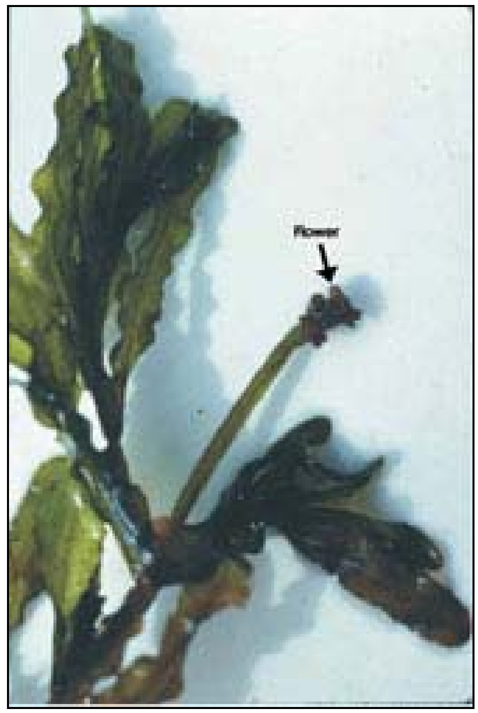
Control – Two biological control agents have been released in southern states – a South American weevil (*Neohydronomus affinis*) and an Asian moth (*Spodoptera pectinicornis*). *N. affinis* successfully established and spread from initial release sites in Florida and Louisiana. Local reductions in water lettuce infestations were observed, but no long-term suppression of infestations has occurred over large areas. *S. pectinicornis* failed to persist at any of the release sites in Florida. Large infestations can be controlled by treating with diquat (Reward®) or glyphosate (Rodeo®) herbicides. Diquat is a non-selective contact herbicide that can be applied throughout the growing season, but is most effective on early growth. Because it is not translocated to all plant tissues, repeated treatments may be necessary. Glyphosate is a non-selective systemic herbicide that is translocated to the root system and may offer more thorough control. Large infestations should be treated in sections, waiting two weeks between treatments to allow time for decomposition. Treating an entire infestation in a single application can result in a significant decrease of dissolved oxygen as the plant matter decays, which can be detrimental to fish and other organisms.

Waterlettuce infestation. Photo by Ken A. Langeland, University of Florida, Bugwood.org.

Compiled from the [Invasive Plant Atlas of New England](), Van Driesche et al. 2002, Florida Exotic Pest Plant Council [Fact Sheet](), and U.S. Army Corps of Engineers Plant Management Information System (PMIS).

Curly-leaf pondweed (*Potamogeton crispus*)





(top) Asexual structure of curlyleaf pondweed. (above) Leaves and flower stalk. Photos courtesy of [Maryland Department of Natural Resources](#).

Description & Biology – Curly-leaf pondweed is perennial, submerged aquatic plant native to Eurasia. It has flattened reddish-brown stems that are branched and vary in growth habit depending on water depth – plants in shallow water may develop as a stunted rosette of leaves, while plants in deeper water can be several feet long. The elongate, reddish-green leaves grow to three inches long and have finely-toothed, wavy margins that give the plant its name. When plants are near the water surface, inconspicuous flowers are borne on short stalks that extend above the water. Seeds are viable, but of limited importance to the spread and maintenance of populations. Hard, prickly winter buds (called turions) that are comprised of small, modified leaves are an important means of reproduction and dispersal. The turions form before plants die back in mid-summer and then fall to the bottom sediment. A single plant may produce hundreds of turions, which germinate in autumn and begin growth during the winter. Once established, the plants form colonies and initiate new growth each spring from rhizomes. The rhizomes are long, thin, and buff or reddish in color.

Curly-leaf pondweed grows in freshwater lakes, ponds, streams and rivers, as well as in slightly brackish water bodies. It tolerates low light, low water temperatures, and is often found growing in alkaline and nutrient rich waters. Its tolerance for cool waters and low light conditions gives it a competitive advantage, allowing it to begin growth prior to native plants in the spring. It dies off in mid-summer when most native plants are growing and the decaying vegetation results in decreased levels of dissolved oxygen and increased nutrient content, which promotes algal blooms. Because of its early growing season, curly-leaf pondweed may not compete with many native plants. However, dense stands can impede boat access and interfere with recreational activities.

Control – For small patches, manual and mechanical removal (cutting, raking, harvesting) initiated in the spring will remove curly-leaf pondweed stands. Removing the entire plant at the sediment surface will discourage production of turions, which should be a main focus of a management plan. All plant fragments must be removed, as any fragment can sprout to form a new plant. Curly-leaf pondweed can be controlled using several herbicides, including endothall (Aquatall K®), diquat (Reward®), or fluridone (®). Applications are best performed in spring and early summer prior to emergence of native plants when temperatures are still adequately low for endothal to be effective. Endothall and diquat are better used for small areas, and generally begin to control curly-leaf pondweed within two weeks. Fluridone is better for large areas, such as an entire lake, and requires 30 days of exposure to be effective.


Dense curlyleaf pondweed growth. Photo by Virginia Kline, University of Wisconsin-Madison, Department of Botany.

Compiled from the Invasive Plant Atlas of New England, Global Invasive Species Database, Wisconsin Department of Natural Resources, and U.S. Army Corps of Engineers Plant Management Information System (PMIS).

Giant salvinia (*Salvinia molesta*)

(top) Close up of "egg beater" hairs on leaf surface. Photo by Mic Julien, Commonwealth Scientific and Industrial Research Organization, Bugwood.org. (above) Typical folding and stacking of densely growing leaves. Photos by Troy Evans, Bugwood.org.

Description & Biology – Giant salvinia, also called water fern, floating fern, water spangles, and kariba-weed, is an aggressive free-floating aquatic fern native to southeastern Brazil that was introduced to the United States for ornamental use. It is federally listed as a noxious weed in the United States and is prohibited by law. A single plant consists of a colony of ramets, with each ramet consisting of a node, an internode, modified submerged leaves ("roots"), and a pair of emergent leaves. The leaves (or fronds) can vary in color from green to golden brown and are densely covered in hairs – each hair is comprised of four cylindrical branches that join at the tip to form an "egg beater" shape, which repels water. It has three distinct growth forms that occur in relation to the age, density of plants, and availability of nutrients. The initial stage of an invasion exhibits small, oval-shaped leaves that lay flat on the water surface. The secondary form has longer internodes and larger leaves that are slightly keeled down the middle and curved along the edge, but still rest on the water surface. Mature infestations form crowded mats with short internodes that press the leaves upright and stack adjacent leaves into long chains. The brown, feathery, submerged leaves appear and function much like roots. Nestled amongst the submerged "roots," round spore-producing structures (called sporocarps) are borne on short stalks, but giant salvinia populations in the United States are sterile and do not produce spores. The only reproduction is through fragmentation of plants and vegetative sprouting. Any plant fragment containing an axillary bud can grow to form a new plant. Humans (boats, ornamental water gardens), animals, and water can disperse plant fragments to cause new infestations.

Giant salvinia typically grows in ponds, lakes, marshes, ditches and other calm water bodies of warm climates. It has been reported in most southern states and as far north as Virginia and Washington, D.C. It exhibits aggressive growth that can blanket entire water bodies. Under favorable conditions, giant salvinia is capable of doubling its population size in seven to ten days. As mature infestations continue to grow, they form a thick multi-layered mat of vegetation that has been observed to grow two feet or more in thickness. Infestations alter aquatic systems in a variety of ways. Dense mats displace native plant and animal life by blocking sunlight and reducing open water habitat. They alter the chemical composition of the water beneath by reducing dissolved oxygen, increasing carbon dioxide, lowering the pH, increasing the temperature, and reducing the availability of nutrients for other organisms. Mats also provide breeding habitat for certain mosquito species that can be disease vectors. In addition, the dense mats impede boat access and recreational activities, and clog irrigation and industrial channels.

Control – Small populations of giant salvinia can be removed manually, dried, and then disposed of in a trash bag. Extensive infestations can be treated with certain aquatic herbicides such diquat (Reward®) and fluridone (Sonar AS®). Diquat may be used in combination with complexed copper (Nautique®) or fluridone. It is recommended that large mats be treated in sections to avoid low dissolved oxygen levels that are associated with decomposing vegetation. Several biocontrol agents have shown promise for controlling extensive infestations, including two salvinia weevils (*Cyrtobaous salviniae* and *C. singularis*) and the waterlettuce moth (*Samea multiplicalis*). Releases of *C. salviniae* in several countries have proven effective at controlling giant salvinia infestations. Weevil populations were found on *Salvinia* species in Florida. Collections were made from these populations and were released in giant salvinia infestations in areas of Texas and eastern Louisiana. These releases are still being monitored to note the long-term effectiveness of control.

Various leaf forms and colors of giant salvinia. Photo by Scott Bauer, USDA Agricultural Research Service, Bugwood.org.

NOTE: Common salvinia (*Salvinia minima*), a native species, looks quite similar to giant salvinia (*S. molesta*), but can be distinguished by its leaf hairs, which do not join at the tip to form the egg beater shape characteristic of giant salvinia.

Compiled from Van Driesche et al. 2002, Invasive Plant Atlas of New England, Plant Invaders of Mid-Atlantic Natural Areas, APHIS Pest Alert, and U.S. Army Corps of Engineers Plant Management Information System (PMIS).

Water chestnut (*Trapa nutans*)

Description & Biology – Water chestnut is an annual, herbaceous aquatic plant native to Eurasia that has invaded aquatic habitats in many northeastern states. It roots in the bottom sediment of water bodies and produces a stalk extending to the surface, which may reach up to 15 feet in length. A floating rosette of leaves is borne at the end of the stalk. The green leaves are triangular or diamond-shaped with coarsely toothed margins. The upper leaf surface is glossy and the lower surface is covered in coarse hairs with obvious veins. The bulbous, spongy leaf petioles allow the leaf rosettes to float. Inconspicuous white flowers are produced in leaf axils at the center of the rosette from July until the first frost. A woody, nut-like fruit is produced underwater that contains a single seed and bears four stout, sharp spines. The fruit sinks to the bottom of water bodies where it may remain viable for up to 12 years, though most germinate within a few years. Each seed can develop into 10 to 15 rosettes, which in turn may produce up to 20 seeds each. Fruits are edible and are dispersed by animals and water movement. Entire or partial plants that have been uprooted may be dispersed to new locations by water currents or human movement.

(top) Water chestnut flower and leaves. (above) Floating mat of leaves. Photos by Leslie J. Mehrhoff, University of Connecticut, Bugwood.org.

Water chestnut grows in ponds, lakes, canals and slow-moving rivers in water up to 15 feet deep, though it prefers shallow water. It grows best in nutrient rich waters that are slightly alkaline. It can form dense surface mats that dominate sunlight, nutrients, and space. Infestations displace native aquatic vegetation, offer little value to wildlife, and reduce dissolved oxygen levels as they decompose. The thick mats preclude recreational activities, such as fishing and swimming, limit boat access, and the spiny fruits can be a hazard.

Control – Various manual and mechanical methods can be employed to control water chestnut. Small populations can be removed by hand, ensuring that the entire plant is removed. Large infestations can be removed or reduced by mechanical means such as harvesters, rotovation machines, and other similar aquatic machinery. The herbicide 2,4-D (AquaKleen® or Navigate®) may also be used to control large infestations. It generally achieves results after about two weeks, and is specific to broad leaf species so native monocots will not be affected. No biological control agents suitable for release in the United States have been identified for water chestnut.

(above left) Underside of water chestnut plant with spiny nut-like fruits. Photo by Alfred Cofrancesco, U.S. Army Corps of Engineers, Bugwood.org. (above right) Close up of nut-like fruit. Photo by Vic Ramey, University of Florida/IFAS Center for Aquatic and Invasive Plants.

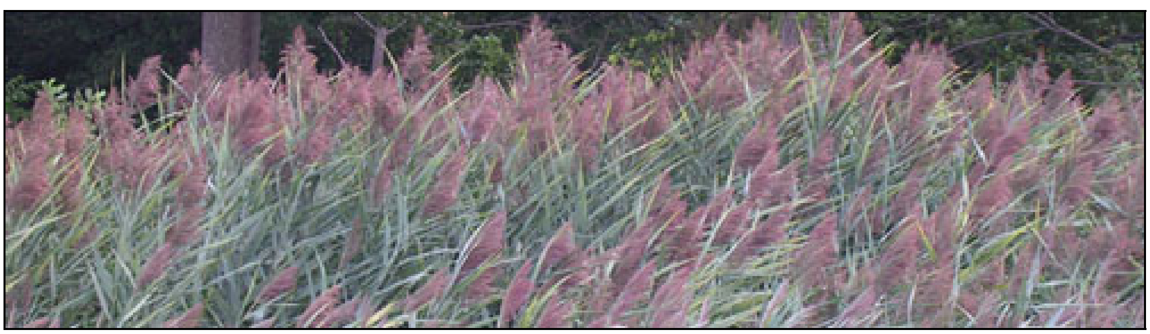
Compiled from Van Driesche et al. 2002, Invasive Plant Atlas of New England, Plant Invaders of Mid-Atlantic Natural Areas, and U.S. Army Corps of Engineers Plant Management Information System (PMIS).

For more in depth coverage of water chestnut management, refer to:
Naylor, M. 2003. Water Chestnut in the Chesapeake Bay Watershed: A Regional Management Plan. Maryland Department of Natural Resources.

Section I – B: Management Techniques

The most effective way to control invasive species is to limit or prevent their introduction from the outset. Prevention is the most cost-effective means to reduce invasions, and it avoids the long-term economic, environmental, and social costs associated with established infestations (VISC 2005). However, absolute prevention is difficult given the prevalence of invasive species across the landscape and the various dispersal routes that invasive propagules may follow. Additionally, budget and staffing limitations may dictate that available resources be directed toward projects that focus on the most damaging species with the greatest potential for success. This can result in a slow or inadequate response to new invasions (DiTomaso & Johnson 2006).

Once an invasive species has colonized an area, rapid identification and implementation of management increases the likelihood of successful control and significantly reduces costs (Simberloff 2003). This is especially true for species with high intrinsic growth rates because even a large initial cost for immediate control is worth avoiding the rapid growth in future costs – both economic and ecological – associated with expansion of an invasion (Olson et al. 2002). The number one guiding principle of the National Invasive Species Management Plan is to take action now to reduce the impacts of invasive species (NISC 2001).



Phragmites (*Phragmites australis*) is a highly invasive and persistent wetland species that has formed monotypic stands in many habitats throughout the Delaware River Basin. Early detection and control of phragmites populations can reduce costs associated with management.

To increase the likelihood of success, invasive species issues must be brought to the forefront of environmental planning on military bases. Installations should identify invasive species management as an objective and target in their Environmental Management System (EMS), as well as in the Integrated Natural Resource Management Plan (INRMP). Defense Instruction 4715.3: *Environmental Conservation Program* addresses policies, responsibilities, and procedures related to ecosystem management, biodiversity conservation, and maintenance and restoration of native ecosystems to support the mission of each installation. The Integrated Training Area Management (ITAM) Program, developed by the U.S. Army Environmental Center (USAEC) and the U.S. Army Corps of Engineers Laboratories, can help to facilitate restoration of degraded military lands. The program engages contractors to restore training areas in order to prevent soil erosion, loss of threatened and endangered species habitat, and degradation of land resources for training. Such efforts can help natural areas resist establishment of invasive species (NISC 2001).

This section discusses the various management techniques available for invasive plants, the limitations and benefits of each technique, and criteria that should be considered when choosing the appropriate methods to include in a management plan. Each situation presents a unique combination of site characteristics, management objectives, and logistical and regulatory constraints. No single management approach will be applicable across a given region or even to different areas within an installation. Typically, a management program that integrates several control methods is most effective at achieving control.

Create Management Plan

Prior to implementing control treatments, a management plan should be laid out to guide control activities. Create a management plan that integrates a logical progression of treatment methods to attain management goals, keeping in mind that methods will likely need to be adapted as you implement control actions and monitor species' responses. Begin by setting management objectives for the site (e.g. clear training areas of nuisance species, or increase native plant diversity) or integrate existing military policies and programs into the invasive species management plan. The installation's Integrated Natural Resources Management Plan (INRMP) likely already includes objectives for managing invasive species. Next, identify which invasive species are preventing the objectives from being reached, or have the potential to do so in the future. Once the problematic species are identified, choose a combination of control methods and implementation schedule most suited to the site conditions and available resources (Tu et al. 2001). Areas with the greatest degree of invasion are not necessarily the best place to begin control. Instead, land managers may wish to focus initially on high quality habitat patches that are high risk areas for invasion, and then move toward patches containing significant infestations (Reed 2004).

As part of Legacy project 08-328, the Wildlife Habitat Council developed an invasive species management plan template to assist DOD land managers with identifying invasive species management priorities and laying out a plan. This document is in review and will be available through the Wildlife Habitat Council's website when released.

After creating a plan and initiating management, control areas should be monitored on a regular basis to evaluate the efficacy of treatments. An adaptive management plan can then be altered accordingly to address changing site conditions. Keep in mind that preservation or restoration of native plant communities should be an underlying goal of management (Tu et al. 2001). Reseeding or planting of native species may be required in control areas following treatments; however, in many instances an adequate native seed bank may be present in the soil. Following release from invasive competition these seeds may germinate to revegetate the area, eliminating the need for replanting.

Regardless of the control methods employed, a key factor for long-term management is the seed bank longevity of both the target invasive species and the native plant community (DiTomaso & Johnson 2006). Management of invasive species should never be considered a one-time venture, as most species build a prolific soil seed bank or have hardy root systems capable of sprouting to form new populations in subsequent years. Control treatments, especially hand pulling and non-selective herbicide application, performed alone with no follow-up management actions can actually worsen an infestation. If funding prohibits multi-year invasive species control, it may be prudent to wait until funding is available for long-term management (Murphy et al. 2007).

Manual and Mechanical Control

Manual and mechanical control methods include hand pulling, cutting, digging and girdling. These methods are generally most effective for small, initial invasions, as they are both time- and labor-intensive, but they may also be used in locations where herbicide or other methods are not suitable. Though manual and mechanical techniques are easily planned and implemented, they often need to be repeated several times during a growing season and in successive years to prevent reestablishment of the target species from seed or vegetative sprouting. Combining a manual or mechanical method with other treatment methods can greatly increase the efficacy of control efforts.

Manual Removal

When pulling or digging, care should be taken to limit the amount of soil disturbance and replace soil where it has been overturned. Areas disturbed by manual removal create prime conditions for invasive species to recolonize the area. Limiting the number of people working in an area will also reduce the amount of disturbance caused by trampling. Annual herbaceous plants, young tree saplings or shrubs, and floating aquatic plants are the easiest to pull by hand. Perennial species with deep root systems are difficult to pull and will often resprout from root fragments that are left behind in the soil. When performing manual removal, it is advisable to wear gloves, long sleeves, and pants to prevent skin irritation, as certain plants can exude substances that may irritate the skin and even fragile plants can leave hands raw after pulling for an extended period of time.

A small demonstration site where Japanese honeysuckle has been hand pulled at Fort Belvoir, VA. Note the surrounding infestation. Photo by Adam Gundlach, Wildlife Habitat Council.

Weed wrench being used to remove young Chinese privet. Photo by Chris Evans, River to River CWMA, Bugwood.org.

Pulling efforts can be aided by tools such as the Root Talon or Weed Wrench, which improve leverage and allow the root system to be pulled along with above-ground growth or severed below the root crown. The

Root Talon is a relatively lightweight, inexpensive tool that is effective against small, shallow-rooted saplings, such as tree-of-heaven (*Ailanthus altissima*) or buckthorn (*Rhamnus* spp.). The Weed Wrench is a sturdier tool made from steel that is better suited to plants with larger stems and roots. It comes in four sizes, depending on the task, but its larger size and weight can make it cumbersome to haul to remote locations (Tu et al. 2001).

Aquatic vegetation can also be removed manually (cutting, dragging, raking, pulling) in certain situations, depending on the target species, size of the infestation, and level of control desired. Just as in terrestrial situations, manual removal of aquatic plants is labor intensive and needs to be repeated frequently to remove new growth. Frequency of removal efforts will depend on the ability of the target species to reproduce and grow, as well as the water conditions – eutrophic waters will generally allow for a higher growth rate (Hoyer & Canfield 1997). Manual removal commonly leaves a significant portion of the target weed population intact, and even thorough clearing of above- and below-ground growth can result in limited population reduction the following season (Murphy 1998).

Advantages of manual removal include targeted removal that limits damage to non-target species in the control area, and low cost for supplies. However, these techniques require a large investment of time and labor, are effective only for relatively small areas, and can potentially promote establishment of invasive species through trampling of native plants and soil disturbance (Tu et al. 2001).

Mechanical Removal

For non-woody species, mowing, weed-whacking, and cutting can be used to prevent seed production or remove above ground biomass prior to implementation of other control treatments, such as herbicide treatment or prescribed burning. The timing of cutting or mowing will depend on the biology of the target species and the surrounding native vegetation. Mowing too early or late in the growth cycle of the target plant may negatively impact native vegetation and be ineffective against the target species, as some species can quickly sprout and reach the flower stage again if cut too early in the season. Cutting certain species may promote greater stem density, and spread plant fragments, which can sprout to form new plants. Be sure to understand the reproductive biology of the target plant prior to cutting. Remove all vegetative material from the site for species that can reproduce by fragmentation. For annual weed species, repeated cutting for several years to prevent seed production can eventually control small patches. The number of years will vary depending on the how well established the target species is and how long seeds are able to remain viable in the soil.

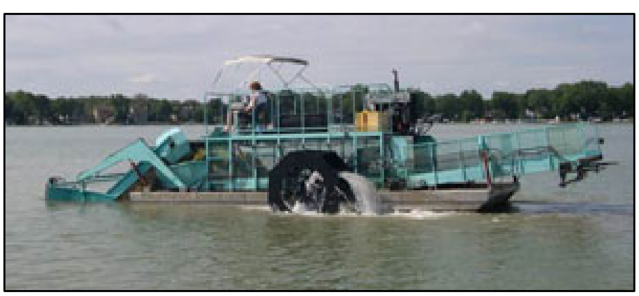
Cogongrass control using mowing. Photo courtesy of USDA APHIS PPQ Archive, Bugwood.org.

Girdling is an effective method to control unwanted trees without completely removing them, but it should only be used for species that do not sprout from the root system. Girdling is accomplished by removing a band of bark from around the circumference of the trunk. Make two parallel cuts around the trunk several inches apart. Be sure to cut deep enough into the bark to sever the vascular cambium (inner bark), which transports nutrients throughout the tree and is the site of new growth. Do not cut too deeply though, as this may reduce the structural integrity of the tree. After completing the cuts, pound on the bark between the cuts with the blunt end of an axe or a similar tool to loosen the bark and peel it away.

Benefits of the girdling method include minimal vegetative material to dispose, little labor input compared with felling and removing trees, increased wildlife habitat for cavity-nesting species – if trees are left standing – and recycling nutrients back into the soil (through decay and decomposition) rather then removing them from the site. Species that should not be girdled include tree-of-heaven (*Ailanthus altissima*), buckthorn (*Rhamnus frangula*), and black locust (*Robinia pseudoacacia*), among others. These species tend to sprout vigorously from the root system following damage, resulting in increased stem density that will require follow-up treatments. Girdling can be combined with herbicide application to increase effectiveness and prevent sprouts from developing. See also the hack-and-squirt method in the next section. Trees should only be left standing following girdling in areas where they will not pose hazard if blown down during high winds (Tu et al. 2001).

Mechanical Removal of Aquatic Species

Aquatic vegetation can be harvested mechanically using specialized machines that either cut or uproot (called rotovation) the plants. This is an important method for control in situations where waterways must be kept clear for boat traffic or recreation because it has immediate results and does not impose restrictions on use, as herbicide application often does. Harvesting also eliminates problems associated with decaying vegetation, such as reduced dissolved oxygen in the water or foul odors, which are common with herbicide application (Hoyer & Canfield 1997). A well-designed mechanical removal program may reduce new growth from perennating tissue by depleting carbohydrate reserves in the root structure of perennial plants. In eutrophic waters, frequent mechanical removal of vegetation may also help reduce nutrient loads (Murphy 1988).



http://aquaticweedharvester.com

In general, the efficacy of mechanical removal of aquatic plants is reduced by lack of uniform coverage in the control area and regrowth of target species (Murphy 1988). Mechanical harvesting is not selective and will remove desirable vegetation along with target species. It creates abundant plant fragments that can drift to new areas to create new infestations. Machines disturb bottom sediment, increasing water turbidity, and also can affect aquatic organisms, such as turtles, snakes, and fish. Mechanical techniques are not feasible in certain water bodies that are too shallow or contain obstructions, and can incur high maintenance and repair costs for machinery (Hoyer & Canfield 1997).

Other manual and mechanical control equipment and tools can be viewed on THE NATURE CONSERVANCY WEBSITE

Herbicides

In 1993, the Secretary of Defense established pest management Measures of Merit (MOM) for DOD, which called for a 50 percent reduction in the amount of pesticide applied on DOD installations by the year 2000 (Parker 1996). In 1996, the U.S. Environmental Protection Agency (EPA) and DOD signed a Memorandum of Understanding (MOU) regarding integrated pest management to reduce risks to humans and the environment associated with pesticides (DENIX 2007). Though efforts such as these have focused on reducing the use of chemical control agents for pest species, in many situations herbicide application is the most effective method to initially contain or reduce invasive plant populations. Perennial invasive species with extensive, vigorous root systems are able to quickly recover to pre-treatment levels following non-chemical treatments, such as pulling, cutting, or burning. Herbicides can kill the roots without causing soil disturbance, which can lead to germination of weed seeds and erosion. Following initial herbicide treatments, more targeted non-chemical techniques can be integrated to eliminate unwanted plants that return to the control area from seeds or root sprouts. Reducing the use of pesticides should be a consideration in any invasive species program, but should not be the sole consideration when choosing a control method. Management decisions should be based on the most effective and environmentally sound control options for the situation, including pesticides (Army Policy Guidance 2001).

Stringent guidelines and regulations are already laid out for proper use of pesticides on military installations, such as DOD Instruction 4150.7 Pest Management Program, which establishes and assigns responsibilities for safe, effective, and environmentally sound integrated pest management programs on military installations (NISC 2001). In light of this, regulatory documents and restrictions related to herbicides are not covered in this publication. This section focuses instead on the methods and equipment used in herbicide treatments.

Considerations

The decision of whether or not to use herbicide in a control program hinges on many factors. Site conditions such as ease of accessibility, proximity to open water, site hydrology, presence of rare, threatened, or endangered species, presence of conservation targets, and the sensitivity of the site to disturbance from application equipment must all be considered. When choosing an herbicide, consider its effectiveness against the target species, its persistence, behavior, and movement in the environment, its toxicity to humans, wildlife, and other organisms, and the mode by which it is applied. Combining multiple herbicides in a mixture can increase the effectiveness and range of susceptible species. Be sure to read all labeling thoroughly before combining herbicides. See Appendix I for a table of commonly used terrestrial herbicides.

The benefits of performing an application should outweigh the potential harm it could cause to the environment. Generally, it is best to use compounds that will not leach into groundwater or spread to nearby surface waters, are not persistent in the environment, and most importantly, are effective against the target species. In certain situations, it may be prudent to use a compound with greater toxicity or persistence if it will reduce the total amount of herbicide needed over the long-term, rather than repeatedly applying a more benign substance (Tu et al. 2001).

Personal protective equipment (PPE) should always be worn by personnel when handling or applying herbicides. Refer to the herbicide label to determine the minimum PPE requirement for the substance being used. Some herbicides are more toxic and require more PPE than others. At a minimum, personnel should wear long-sleeved shirts, pants, sturdy boots (preferably chemical-resistant rubber), safety glasses, and rubbers gloves (tyvek or nitrile are recommended). Certain materials such as cotton, leather, and canvas will absorb chemicals (even dry formulations), are difficult to wash thoroughly, and are not recommended when handling herbicides.

The costs and benefits to wildlife should always be considered when planning herbicide applications. Adding chemicals to the ecosystem can be detrimental to existing species on the site; on the other hand properly implemented herbicide treatment allows for the restoration of native habitat that species need for survival. Photo: eastern swallowtail (*Papilio glaucus*) by Adam Gundlach, Wildlife Habitat Council.

Pesticides and Pollinators

Given recent concerns over Colony Collapse Disorder (CCD) in honey bees throughout world and declining populations in other pollinator species, it is important to consider the impact a proposed herbicide application may have on these ecologically crucial species. The North American Pollinator Protection Campaign (NAPPC) has compiled a list of online resources related to reducing risks to pollinators when using pesticides. The information can be accessed at http://www.nappc.org/PesticidesMain.html. Costs and benefits of herbicide applications must be considered. Adding chemicals to the ecosystem can be detrimental to pollinators currently using the site; on the other hand, invasive plants often contain phytochemicals that severely impact pollinators' reproductive success (e.g. DiTommaso & Losey 2003, Casagrande & Dacey 2007), and sometimes herbicide is the most effective method to remove these invasive species and restore the native plant communities on which pollinators depend. The important thing is to plan herbicide treatments carefully in order to use the right amount of the right chemical in the right place. The EPA's Pesticide Environmental Stewardship Program offers information about reducing risks related to pesticide use.

Herbicide Application Methods and Equipment

The herbicide application method that is most appropriate depends on the target species, the herbicide being used, the size and accessibility of the treatment area, and the available equipment. Techniques described below can be modified to best suit the site conditions.

Boom applicator mounted on an ATV. Photo James H. Miller, USDA Forest Service, Bugwood.org

Foliar Application

Foliar applications refer to methods that apply herbicide directly to the leaves and stems of plants. Techniques for foliar application can target individual plants (spot applications) or can cover large areas in extensive infestations. Spot applications can be performed using vehicles mounted with spray hoses, backpack sprayers, hand-pumped sprayers, squirt bottles, or wick applicators. The most appropriate equipment depends on the size of the target species, its density and proximity to desirable vegetation, and its accessibility. Foliar sprays applied with a back pack sprayer are the most common application method. To limit spray drift to non-target vegetation, herbicide mixtures can include drift retardant compound (though this may reduce herbicide effectiveness), spray shields can be attached to the nozzle, and the herbicide should be applied at low pressure to limit fine particles. Wick applicators use a sponge or other wicking material – often mounted on a hollow handle that holds the herbicide – to wipe herbicide directly on target vegetation, thereby reducing the potential for application to non-target plants. Wide coverage applications can be made with a boom applicator or multiple spray nozzles mounted on an ATV, tractor (or other vehicle), helicopter, or small plane. Such applications increase the chance of herbicide drift to non-target areas, but allows for large infestations to be treated efficiently (Tu et al. 2001, Miller 2003).

Backpack sprayer. Photo by Chris Evans, River to River CWMA, Bugwood.org.

Widecast application to an extensive kudzu infestation. Photo by James H. Miller, USDA Forest Service, Bugwood.org

Adjuvants, most importantly surfactants, often need to be included in the herbicide mix (refer to herbicide labeling) in order to penetrate the waxy cuticle layer of leaves and stems, which can prevent uptake of herbicide into plant tissue. Though water is the most common and economical carrier for herbicide mixtures, other substances can also be used as carriers with many herbicides, and some may reduce the amount of herbicide needed per acre. Thinvert® is a formulation of non-phtyotoxic paraffinic oils, surfactants, and emulsifiers that forms a thin invert emulsion. It can be used as a carrier in place of water and is applied with a specially designed application system that produces uniform droplet size. The uniform spray pattern and white coloration of the substance allows the applicator greater visibility and control. Thinvert mixtures reduce spray drift and decrease evaporation associated with movement through the air. The oily nature of the mixture and low application volume prevents runoff and ensures retention on foliage, which can increase herbicide uptake by the plant. Thinvert's combination of properties allows it to be an effective carrier at application rates as low as five gallons per acre (Gover et al. 2003).

Basal Bark Application
This method involves applying an herbicide-oil-penetrant mixture to the lower 12 to 20 inches around the entire trunk/stem of woody vegetation. Certain herbicides, such as Pathfinder® II and Pathway®, are sold pre-mixed and ready-to-use with the appropriate ingredients. Basal applications can be sprayed, applied with a wick applicator, or painted on with a brush to the target plants. Ester formulations of herbicide are generally most effective for basal bark treatments because they penetrate bark easier than salt formulations. Ester formulations should be applied on calm, cool days due to their volatility. Basal bark applications are generally more effective against young trees with smooth bark, as the thick furrowed bark of mature trees often prevents adequate uptake of herbicide (Tu et al. 2001, Miller 2003)

Hack and squirt method. Photos by Steve Dewey, Utah State University, Bugwood.org.

Cut-stump Application
This selective method is typically used for woody species that tend to resprout following damage. It involves cutting trunks/stems near or just above ground level and immediately applying an herbicide concentrate or mixture to the freshly cut stump. Herbicide can be applied with a sprayer, spray bottle, wick applicator, or paint brush. Small stumps should be completely coated with herbicide, while the outer circumference of larger stumps can be treated in a three-inch band, where the actively growing tissue is located. Avoid applying herbicide to the point of runoff. Remove sawdust from the stump prior to applying the herbicide, as the sawdust can serve to deactivate the herbicide before it

is absorbed. Stumps that have remained untreated for two hours or more should be treated an herbicide-oil-penetrant mixture, as described in "Basal Bark Application" above, rather than a standard water-based mixture. Woody vegetation treated with a cut-stump treatment may still resprout and should be monitored regularly and retreated as needed (Tu et al. 2001, Miller 2003).

Injection and Frill Application

Hypo-Hatchet® showing hole in bit where herbicide is injected. Photo by Gerald J. Lenhard, Bugwood.org.

These selective methods target larger trees and shrubs without damaging surrounding vegetation. Frill applications, also called the hack and squirt method, are made by using a hatchet, ax, or other implement to make downward cuts through the outer bark spaced around the circumference of a tree. The cuts should be treated immediately with herbicide applied with a sprayer, squirt bottle, or syringe. Avoid runoff from cuts if possible. Soil-active herbicides should not be used in areas with desirable vegetation, as the herbicide can be leached to the soil through the roots of the treated plant. Specialized tree injector tools are also available. These typically consist of a metal shaft with sharp metal point that is thrust into the tree to create an incision. A lever on the device is then pulled to release herbicide into the cut in one step. Other tree injectors insert measured capsules of herbicide into the tree upon impact (E-Z-Ject® Lance). The Hypo-Hatchet® is hatchet with an herbicide reservoir in the bit that injects a standard amount of herbicide upon impact. Avoid using injection or hack and squirt treatments during spring growth because the heavy sap flow during this period can flush herbicide from the cuts and limit effectiveness (Tu et al. 2001, Miller 2003).

Aquatic Herbicides

Herbicides labeled for aquatic use fall into two categories either "contact" or "systemic." Contact herbicides are fast acting and generally cause cell damage to any tissue that comes in contact with the herbicide, mainly stems and foliage. Unfortunately, their effects are not sustained and often fail to reach perennial tissue such as the roots, root crowns, and rhizomes. Systemic herbicides are absorbed and transported to all plant tissues, resulting in a more complete kill. However, systemic herbicides are slower acting and must remain in contact with target vegetation for longer periods to be successful. The extended exposure time required by many systemic herbicides preclude their use in wetland systems with high flow rates. See Appendix II for a table of commonly used aquatic herbicides.

Herbicide Disposal and Reuse

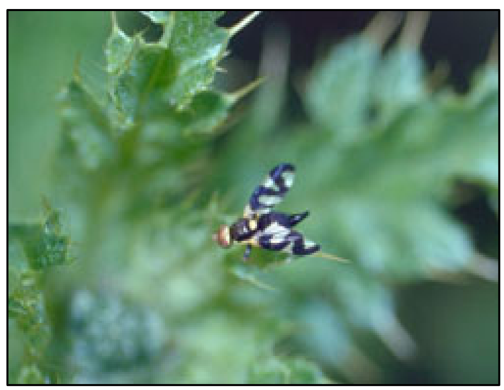
The Department of Defense Resource Recovery and Recycling Program (RRRP) aids in reutilization of excess equipment, products, and materials that are no longer needed by the original proprietary installations or military services. Excess property may be turned into the Defense Reutilization and Marketing Service (DRMS) where it is then redistributed, transferred to other Federal agencies, or donated to other state and local government agencies. In fiscal year 2006, $1.9 billion worth of surplus property was reutilized. DRMS manages the disposal or reuse of hazardous properties, including herbicide, minimizing the environmental risks and costs associated with disposal. Installations that have excess herbicide or equipment can transfer it to DRMS so it may be redistributed to other installations or properly disposed.

Biological Control

Biological control (biocontrol) is a scientific method for identifying natural control agents of invasive species and introducing the agents into the invaded range or promoting existing agents (Van Driesche et al. 2002). Biocontrol agents can include animals, insects, fungi, or microorganisms that interfere with the ability of a pest species to cause damage or persist in the landscape. Despite the potential for biocontrol agents to negatively impact native vegetation, they represent one of the most important and promising methods for controlling invasive species over large areas.

Biocontrol is generally viewed as an environmentally sound control method because it avoids use of chemical compounds that can harm humans and non-target organisms. It can be effective for reducing invasive species across large areas for extended periods of time with minimal input of labor and funding. There are three categories of biocontrol. The first and most widely used is 'Classical' biocontrol, which employs control agents from a non-native pest's home range. 'Neoclassical' or 'New Association' biocontrol pairs non-native control agents against native pest species. Lastly, 'Conservation', 'Augmentation', or 'Inundation' methods aim to promote populations or increase the impact of pre-existing control agents, which are often native to the area (Tu et al. 2001).

Classical biocontrol assumes that invasive species have escaped the natural enemies that exist in their native range, and aims to reconnect the target invasive species with one or more of these natural enemies. Classical biocontrol efforts can reduce the abundance or impact of target species across large areas with minimal damage to other vegetation. Certain control agents fail to establish or require repeated releases before establishing sustainable populations. Once established, the control agent theoretically offers perpetual control without further input of resources, and can even spread to new locations. This trait can become a liability if the control agent has unintended affects on non-target species.



Canada thistle stem gall fly (*Urophora cardui*) and galls. Photos by Alec McClay, McClay Ecoscience, Bugwood.org

Conservation biocontrol generally preserves or promotes existing organisms (native and non-native) that could potentially control populations of pest species. This technique has received relatively little attention or study. Native control agents will only be effective against a handful of invasive species, but they are less likely to cause damage desirable species than non-native agents.

Inundation or **augmentation biocontrol** uses mass releases to overwhelm target species with control agents that are generally already present, but not in adequate numbers to achieve large-scale control. This technique is most often used against insect pests, but agents such as grass carp have been used against aquatic vegetation. When agents that are not host-specific are used, they can be sterilized to avoid establishment and long-term damage to non-target organisms. Inundation agents often need to be released multiple times because they fail to establish viable populations or persist at levels that are adequate for controlling the target species over the long-term.

New association or **neoclassical biocontrol** employs non-native control agents against native pest species, typically ones that are exceptionally dominant in their range or that cause significant economic losses. Ideally, the control agent is host-specific and capable of reducing the native pest to acceptable levels without completely eliminating it. Targeting native species with non-native control agents is risky though, considering that the control agents have no evolutionary connection with the target species, and after being released, could find any number of other non-target species just as suitable (Simerbloff & Stiling 1996). The upside of a successful new association control program is the resultant decrease in pesticide use and labor associated with management activities. However, like classical biocontrol, the control agents released are not native and have the potential to adapt to the new environmental conditions and damage non-target species (Tu et al. 2001).

Alligatorweed flea beetle (*Agasicles hygrophila)* larvae and adult. Photo by Gary Buckingham, USDA Agricultural Research Service, Bugwood.org.

Loosestrife leaf beetle (*Galerucella calmariensis*) larvae and damage to foliage. Photo by Linda Wilson, University of Idaho, Bugwood.og.

Selection of biocontrol agents requires a significant period of time prior to release to screen the agents for host-specificity, which helps to avoid negative impacts to non-target species. Once the screening process is completed, biocontrol programs require little more than propagating or obtaining control agents and releasing them into the appropriate areas. Biocontrol programs can take several years before showing signs of success, as the control agents may take this long to establish populations sufficient to inflict damage on the target species. Releasing multiple control agents against a target species can result in competitive interference, and may impede a control agent that would otherwise be effective (Ehler & Hall 1982 in Denoth et al. 2002). However, biocontrol programs that use multiple control agents against invasive vegetation often realize increased effectiveness to a certain point. Competitive interactions are not always directly related to the number of control agents released (Denoth et al. 2002), but competition is more likely with greater numbers of control agents, as are negative impacts to non-target species. Biocontrol programs should focus on identifying species that are most likely to be effective, and release the minimum number of control agents necessary for successful control of the target species (McEvoy & Coombs 2000 in Tu et al. 2001).

Biocontrol will never completely eradicate a target species, but can reduce the population to acceptable levels or mitigate damages (Tu et al. 2001). Regardless of the extent of screening performed, releasing biocontrol agents will always pose inherent risks to non-target species. Unlike other control methods, biocontrol agents can disperse and adapt to their new environmental conditions, making it difficult to predict with certainty the total impact of the introduction (Simberloff & Stiling 1996). Even biocontrol agents that remain specific to the target host and do not disperse widely can have unforeseen consequences when they harbor other parasites or pathogens.

The benefits and risks of biocontrol should be weighed against those of other control methods or absence of control altogether. In cases of extensive infestations where an invasive species has significantly altered an ecosystem by excluding native vegetation, the risks of a well planned biocontrol program will likely be minimal compared with continued degradation and reduction in biodiversity caused by the invasive species. Before implementing a biocontrol program for invasive species, consult with appropriate local, state, and federal agencies regarding the proposed control agents to ensure their efficacy and to avoid potential detrimental affects to the ecosystem (Army Policy Guidance DAIM-ED-N 2001).

Prescribed Fire

Prescribed fire, also called prescribed burning, can be used in certain situations to achieve various management goals, and when incorporated into an installation's Integrated Natural Resources Management Plan (INRMP) or Integrated Training Area Management Program (ITAM), can be a valuable tool for managing invasive species in a cost effective manner. Prescribed fire can serve multiple functions by restoring natural disturbance regimes and reducing hazardous fuel loads, while also controlling invasive species populations and promoting native

Photo by Wayne Adkins, USDA Forest Service, Bugwood.org.

plant communities. The use of fire as a management tool also presents several issues that must be addressed in the planning and implementation phases of a management program (DiTomaso & Johnson 2006).

Decades of fire suppression has altered historic wildfire patterns and converted fire-adapted communities, such as pinyon-juniper, chaparral, and ponderosa pine in the western United States and long-leaf pine, prairie, savanna, and other grassland communities in the eastern U.S., to fire-prone, late-successional ecosystems (CEMML, DODWFMPWG). Frequent wildfires are a natural and often crucial component of many ecosystems throughout the U.S. It has been suggested that fire suppression has contributed to habitat degradation for numerous plant and animal species and increased the risk of catastrophic fires through increased fuel loads. Consequently, the frequency of severe wildfires has increased dramatically in recent years, especially in the western U.S., but also in the eastern U.S., as evidenced by the large wildfires along the Florida-Georgia border in the spring of 2007. Wildfire is a real threat to military installations, as many installations are bordered or surrounded by lands that support fire-adapted vegetative communities. Installations where live ammunition training occurs have an even greater risk of fire (U.S. E.P.A. 1992). Other installations have become islands of biodiversity in the urban matrix, increasing the risk of wildfire escaping from an installation into surrounding urban development (DODWFMPWG). Prescribed fire allows land managers to control the timing and intensity of fires to reduce fuel loads safely while maximizing the benefit to ecosystems and reducing the cost and impact to human environs – including military installations – associated with wildfires (CEMML, ColoState).

Controlling Invasive Species with Fire

As a management tool, prescribed fire can be used to control invasive plants, increase native plant populations, and improve wildlife habitat. Fire can be effective against annual, perennial, and woody invasive plants through direct damage and suppression of seed production in the target species. Though many examples exist of successful control using fire, relatively few invasive species have been evaluated for their suitability to such programs. Long-term control of invasive species requires prevention of all reproductive structures, which fire alone may not be able to accomplish. When incorporated into an integrated management program, fire can enhance the effectiveness of mechanical, chemical, or cultural control methods (DiTomaso & Johnson 2006).

Photo by Dale Wade, Rx Fire Doctor, Bugwood.org.

The weather conditions, topography, and fuel load of the habitat will influence the intensity and duration of the burn. These factors along with the timing of the burn will influence the impact on target species and native plant communities. Generally, prescribed fires are most effective when performed just prior to flower or seed production for herbaceous species, or during the seedling/sapling stage for woody species (Tu et al. 2001).

Burns to control annual species must be initiated before viable seeds are produced or before seed dispersal occurs. Ideally, a burn should be conducted while seeds of the target species are still in the burn canopy – where the highest temperatures are reached – and after the seeds of desirable species have dispersed to the ground. Seeds that have been dispersed to the ground will not be affected by most burns because lethal temperatures are not reached at ground level unless there is high fuel loads present (DiTomaso et al. 2006). Not all annual species respond the same to fire, and responses may vary due to the timing of the burn and seed moisture content (DiTomaso & Johnson 2006).

Biennial species can be difficult to control with fire because they exist in mixed-age stands, and first-year plants in the rosette stage have protected meristems that will not be damaged by most burns. Second-year plants are susceptible to fire after bolting, but the burns must be conducted prior to seed set. Higher intensity fires exhibit greater control of biennial species. Single burn events do not control biennial species. Burning for consecutive years can effectively decrease the seed bank and prevent new seed production, but may not be viable in habitats where a sufficient thatch layer does not accumulate between burns. Using prescribed fire in combination with timely herbicide treatments will likely be most effective against biennial species (DiTomaso & Johnson 2006).

A successful management strategy for perennial species must be long-term. In regions that formerly supported prairie or grassland habitat, prescribed fires can increase the abundance and diversity of native warm season species and suppress perennial non-native cool season grasses. Fires should be conducted in the spring when cool season grasses are elongating and native warm season plants are still dormant. A thick thatch layer is also needed for adequate control of the cool season grasses. Frequent spring burns increase the suppression of non-native grasses (Rice 2005). If warm season natives are not present in the burn area, the non-native cool season grasses will likely re-establish without further management (DiTomaso & Johnson 2006).

Perennial forbs and woody invasive species are difficult to control with fire. Fire actually promotes many species, particularly species that sprout vigorously following fire damage or possess seeds that are specially adapted to germinate following a burn. Effective management often requires integration of other control methods, particularly herbicide (DiTomaso & Johnson 2006, Tu et al. 2001). Burns performed to control invasive woody vine species are rarely successful. However, in areas where perennial vines remain evergreen or semi-evergreen, burning during the winter can reduce the abundance or stop the spread of invasive vines with little damage to native plants (Tu et al. 2001). Tree seedlings and saplings are most susceptible to fire. Larger trees can be killed given a sufficiently hot fire, but many species will resprout after being top-killed. Higher fuel loads can be achieved by cutting underbrush earlier in the season and allowing it to dry prior to a burn. Other management programs have increased fuel loads by seeding an annual grass species below target woody vegetation prior to burning. In the Mid-Atlantic, burns performed against perennial woody species during the dormant season are generally unsuccessful because of resprouting that occurs the following growing season. Burning during the growing season may improve results against woody species because this is when carbohydrate reserves are lowest (Richburg & Paterson 2003 in Ditomaso et al. 2006), and depletion of energy reserves in the roots may make plants more susceptible to ensuing treatments. Repeated burns will likely increase suppression of woody species, but may not be practical in all areas.

Burning with a Propane Torch

Small, localized patches of invasive plants or scattered individuals may be spot-burned using a propane torch. Propane torches allow for highly selective control that avoids desirable vegetation and is effective against the target plants. Propane torches are also useful for follow-up spot treatments on individual plants, including first-year rosettes of biennial species, after performing a prescribed burn or herbicide application. It can also be used in damp or otherwise inadequate conditions that would prevent a traditional fire from carrying. Like with other methods, repeated treatments are often necessary to eliminate sprouts that develop from seeds and root structures (Tu et al. 2001).

Integrating Fire and Other Control Methods

Few invasive species can be controlled with a single burn treatment. In many situations repeated burns are impractical, but even where multiple-year burns are possible, they may not be suited to the habitat and other control methods will need to be worked into the management strategy to effectively control most invasive species. In some areas, prescribed fire may not be allowed at all, but the affects of a naturally occurring wildfire could be incorporated into invasive species management (DiTomaso & Johnson 2006). Fire will often promote germination of native seeds, but certain invasive species are actually well adapted to fire as well, and may flourish after a burn. When this occurs, other control methods, such as herbicide application, will need to be integrated into the management strategy to squelch the abundant sprouts and seedlings that often develop following a burn (Tu et al. 2001).

Prescribed fire being used to remove the thatch layer prior to herbicide applications for cogongrass. Photo by David J. Moorhead, University of Georgia, Bugwood.org.

An initial burn can stimulate seed germination and deplete the seed bank of a target species. The following year's seedlings can then be treated with an herbicide application or mechanical treatment (DiTomaso et al. 2006). Prescribed fire also helps to prepare a site for subsequent treatments by clearing the area of vegetative debris and revealing hazards of the topography (Miller 2003). This allows easier access for machines used in mechanical control and herbicide application. Removal of the thatch layer, or leaf litter, improves deposition of pre-emergence herbicides, increasing soil penetration and subsequent uptake by the roots (Winter 1993 in DiTomaso & Johnson

2006). Removal of the thatch layer also increases contact of foliar herbicide sprays with leaves of young sprouts, providing for better control.

In certain situations, prescribed fire is better used as a follow-up to other treatments. Herbicide treatments create dead vegetation that can enhance the intensity and ability of a fire to carry (Glass 1991 in DiTomaso et al. 2006). An herbicide or mechanical treatment may also encourage growth of native grasses or understory species, providing fuel to carry a later burn. In other cases, prescribed fire may be used between treatments to remove dead biomass and promote growth of the target species before a second treatment. This method has been used successfully to control common reed (*Phragmites australis*), and exhibited greater effectiveness than treatments relying solely on herbicide (Clark 1998 in DiTomaso & Johnson 2006).

Prescribed fire can kill biological control agents, especially control agents with larvae that feed in the seed or flowerheads of target species. However, if burns are timed to coincide with dormant periods of a control agent's life cycle, negative impacts can be averted. Even when burns negatively affect biocontrol agent populations, a population is often able to rebound relatively quickly through immigration from outside of the burn area (DiTomaso & Johnson 2006).

With an integrated approach, a management program can be tailored to select for a more desirable plant community and prevent a single species from dominating burned areas. Species that complete their life cycle before a burn will generally be selected for, and species that set seed after the time of the burn will be negatively impacted. Integrated management may also reduce dependence on herbicide by increasing its efficacy and reducing the number of treatments required for control (DiTomaso et al. 2006). Prescribed burns can aid in revegetation projects by clearing an area of dense thatch, increasing soil temperatures to promote germination of native seeds, and eliminating invasive species, which would otherwise compete with desirable vegetation. This gives reseeded native species a window of opportunity to become established and help to suppress invasive species. Reseeding can also be used to prevent erosion following a burn (DiTomaso & Johnson 2006).

Planning a Burn

Planning and conducting a prescribed burn often involves input and participation from a variety of agencies and stakeholders. One organization or individual, often called the lead agency, should be designated as the responsible party for the project. The lead agency should set management objectives and review all the factors involved to determine if a prescribed burn is the appropriate tool for the project, given the associated risks. The lead agency must also ensure that all regulatory requirements are met and assume full legal

A fire crew watches over a burn to control diffuse knapweed (*Centaurea diffusa*). Photo by Steve Dewey, Utah State University, Bugwood.org.

liability for the project, or negotiate the terms of liability with other participating agencies and organizations (DiTomaso & Johnson 2006).

A coordinator should be appointed to develop a burn plan and gain approval for the project. The burn plan should contain goals that move site conditions toward the stated objectives, or the objectives contained in an installation's INRMP. The plan should include background information on the site, justification for the proposed burn, management goals for the burn, a proposed fire regime, and specific guidelines for conducting the burn – including acceptable environmental conditions (humidity, temperature, soil moisture, topography, fuel load), equipment needed, potential sources for emergency assistance, smoke management guidelines, considerations for the local community, maps of the burn area, and a checklist to use in preparation for the burn (Tu et al. 2001). Many environmental factors are often overlooked when developing the burn plan, which may account for the variability in results exhibited by prescribed burns. More research is needed into how these factors affect the success or failure of a burn, so that land managers may more accurately predict the outcome of a burn under specific conditions (DiTomaso & Johnson 2006).

The plan may need to be reviewed by various government agencies and private organizations that will approach the project from opposite perspectives. Prescribed fire projects often garner public complaints and obstacles, which arise from a poor understanding of the problem, disagreement with the proposed method, or distrust for the organization(s) proposing the action. Patience and perseverance will likely be needed to gain approval from all stakeholders for certain projects (DiTomaso & Johnson 2006).

Once approved, a qualified fire manager should execute the burn plan with qualified personnel. The fire manager must confirm that all equipment is in place, weather conditions are acceptable, and the appropriate agencies and authorities have been notified to ensure the burn is conducted in a safe manner. The ideal time to initiate a burn may coincide with a time when adequate resources are not available to execute the burn properly because of other wildfires, burn projects, or limited funding (DiTomaso & Johnson 2006). In such instances, the best and safest option may be to take no action rather than implementing a burn with insufficient personnel or equipment. The benefits of the carrying out the project should outweigh the overall risks and costs. After a prescribed burn is performed, the burn area should be monitored to assess the impacts to target and non-target species (Tu et al. 2001).

Effects of Fire on Native Plant Communities and Soil

Fire is a complex management tool with myriad aspects and effects on an ecosystem, and variable outcomes. Not all burns or burn cycles (regimes) are beneficial, and some may even decrease the abundance and diversity of native species. Burns that are too hot can damage the root crown of native plants, kill seeds and microorganisms in the soil, or alter the chemical composition of the soil by destroying nutrients (Tu et al. 2001). Burning too frequently can create an unnatural fire regime that may favor non-native species, as most native ecosystems did not evolve with short fire interval. Disturbance associated with repeated burns can result in bare ground being exposed for extended periods, increasing the risk of soil erosion, especially on steep topography (Brooks et al. 2004 in DiTomaso & Johnson 2006).

One of the most significant gaps in knowledge about prescribed fire is how it affects native vegetation. Given the range of environmental conditions that can exist before and after a burn, the complex interactions between members of a plant community, and the varying characteristics (fuel consumption pattern, fire intensity, speed) and impacts of a burn on individual species, it is often difficult to predict the resultant plant community. Invasive species are generally well adapted to disturbed habitats and tend to dominate post-fire habitats unless native fire-adapted species are also present. If native plant populations are inadequate, reseeding may need to take place immediately following a burn to prevent reinvasion of the treatment area (DiTomaso & Johnson 2006).

Typically, only high temperature fires negatively impact the chemical, biological, and physical properties of soil. Such fires tend to occur in areas where fire suppression has created unnaturally high fuel loads or in highly productive forest and shrub communities. Slow-moving, smoldering fires result in greater soil heating, and moist soils conduct heat deeper below the surface than dry soils. Managing for fast-moving fires conducted under dry soil conditions will limit negative impacts to soil properties. Nutrients in the soil volatilize at different temperatures. Organic matter and nitrogen can volatilize at relatively low temperatures around 200°C. Other nutrients, such as potassium, phosphorous, calcium, magnesium, and sodium, require significantly higher temperatures (700°C or greater) for volatilization, and should not be affected by most fires. The pH of the upper layer of soil will typically increase following a fire (Neary et al. 1999), but will quickly decrease once plants begin to grow again.

Microorganisms, such as bacteria and fungi, play a key role in making soil nutrients available for uptake by plants. Microbes at the surface can be killed when temperatures exceed 100°C, but the insulative quality of soil protects organisms in deeper soil from all but the hottest fires. Mycorrhizal fungi play an important role in a plant's ability to obtain water and nutrients from the soil. Typically, fires do not significantly alter mycorrhizal fungi populations unless they are exceedingly hot or prolonged. A reduction in mycorrhizal fungi can result in poor establishment of plants following fire. If fungi populations are reduced, they will recover over time through migration from deeper soil and dispersal of fungal spores (DiTomaso & Johnson 2006).

Cattle grazing on kudzu. Photo courtesy of USDA NRCS Archive, Bugwood.org.

Grazing

In some situations, grazing can be incorporated into a management program to reduce infestations of certain invasive plants. A properly run grazing program, when combined with other control methods, can completely eliminate small invasions and significantly reduce large invasions. However, improperly managed grazing programs can promote the spread and establishment of invasive species through excessive soil disturbance and over-grazing of native plants. Additionally, moving herds between control areas and pastures can spread seeds of invasive species to new locations, so care should be taken to reduce the likelihood of this occurring. Conversely, soil disturbance caused by a herd may also be used beneficially in restoration efforts to help sow native seeds into the soil while the herd grazes (Tu et al. 2001).

To initiate a grazing program, consider forming a partnership with a local farmer or rancher to temporarily relocate a herd to the control area, or to set up a rotational grazing schedule. A grazing program will often be limited by availability of livestock in the local area, as transportation costs can be prohibitive. Cattle, sheep, goats, and horses are the most common livestock used in grazing programs. Sheep and goats tend to prefer broadleaf herbaceous plants, but goats will eat a broader range of forage material than sheep. Sheep prefer forbs over grasses and grasses over shrubs, whereas goats typically prefer shrubs over forbs or grasses. Depending on the situation, goats and sheep may not be the best choice for management as they can compete with Both sheep and goats are capable of breaking down certain phytochemicals found in some noxious weeds, such as leafy spurge (*Euphorbia esula*) and Russian knapweed (*Acroptilon repens*), though St. Johns Wort (*Hypericum perforatum*) and senecio (*Senecio* spp.) can be toxic to sheep. Another benefit of sheep and goats is that they can traverse steep, rocky terrain, allowing them access to areas that other livestock cannot reach. Cattle prefer grass Horses are more selective than cattle, tending to only eat grasses, so their use will be restricted to infestations of invasive grasses (Tu et al. 2001).

Animals should be introduced into a control area at a time when they will cause the greatest damage to target species and least damage to native species. The optimal time generally occurs during the early flower stage, but timing will also be influenced by the palatability of vegetation, as certain species, such as cheatgrass (*Bromus tectorum*), are only palatable during early stages of growth. A compromise will often need to be made between the most susceptible period to initiate grazing for the target species and the period of greatest palatability. In the case of herbaceous broad-leaf plants, the early rapid growth phase offers livestock the most nutritional value, and coincides with the time of greatest nutritional need for young livestock and mothers. Susceptibility of native plants to grazing should also be considered, as native plants may be most palatable during the same period as the target species, and livestock may prefer the natives, resulting in a competitive disadvantage for the native plants (ASI 2006). One final thing to consider at the end of a grazing cycle is that seeds of invasive species may remain viable after being digested by livestock, so

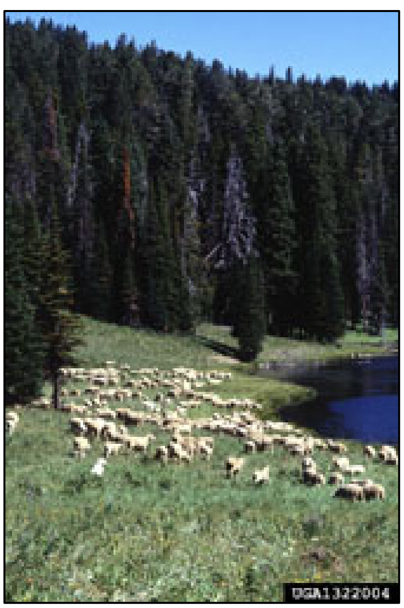


Photo by Scott Bauer, USDA Agricultural Research Service, Bugwood.org.

herds should be kept out of uninfested locations for nine days to prevent dispersal of seeds.

The duration of a grazing program will depend upon the weed species being targeted, the animals being employed, and the native vegetative community present. Initially, the stem density of the target species may increase following grazing, but with repeated defoliation over the long-term, an infestation should be reduced. Often, herds will be confined to a small portion of the control area for a period of time (several days) until desired reductions in the target plant are achieved, or when native plants begin to be negatively impacted. The herd is then moved to another portion and the process repeated. Once the herd has been rotated through the entire control area, a second grazing rotation will often be needed during the same growing season to remove new growth that has occurred. Grazing programs will not result in complete eradication of a target species, but when managed properly for several years, they can reduce weed populations to manageable levels, and when coupled with other management techniques, may achieve complete control of the target species.

Clean Equipment

All equipment used in control efforts, including boots, clothing, tools, and vehicles, should be thoroughly cleaned before being moved to a new site. Equipment contaminated with seeds or vegetative material of invasive species can easily spread propagules to new locations (Tu et al. 2006). Vehicles and machinery should be hosed down thoroughly (preferably pressure washed) to remove any soil that may contain seeds. Runoff from cleaning operations should be contained and prevented from entering streams, rivers, or other waterways that could disseminate seeds or other plant parts.

DOD vehicles and cargo are subject to "Agricultural Cleaning and Inspection Requirements" under DOD 4500.9-R, Defense Transportation Regulation, Part V, Chapter 505. These requirements aim to prevent "any movement that has the potential to introduce invasive species to a new area." Vehicles and equipment of private contractors working on DOD installations are also likely sources for movement of invasive species. These vehicles should be held to the same cleaning and inspection requirements as military vehicles if invasive species prevention is to be successful. Utility vehicles commonly introduce propagules of invasive species along roadways and in construction areas, which offer relatively safe locations for germination and establishment of invasive species (Schmidt 1989, Greenberg et al. 1997, Trombulak & Frissell 2000). Managing pathways of invasion is one of the easiest and most efficient ways to prevent unintentional introductions of invasive species (NISC 2001). As the

A vehicle is pressured washed prior to customs inspection. Photo courtesy of U.S. Army Materiel Command (AMC).

Department of Defense often must accomplish more with less, especially in natural resource management (Goodman 1996), preventative measures, such as stringent vehicle and equipment cleaning, will be important means of avoiding future management costs associated with invasive species introductions.

Refer to *Technical Guide No. 31 – Retrograde Washdowns: Cleaning and Inspection Procedures* produced by the Armed Forces Pest Management Board, which provides information on cleaning techniques and inspection procedures currently used by DOD personnel.

Section II:
Preventing Recurring Invasive Species and Restoring Historical Plant Communities

To be effective, invasive species control must be approached holistically, in the context of a long-term management plan. Treating invasive plants as a maintenance issue to be dealt with on an annual basis will generally only serve to perpetuate the target populations, and may even increase the vigor or spread of certain species. The temporary focus of such an approach only achieves short-term reductions in plant growth that will need to be repeated indefinitely. Traditionally, many land managers controlled invasive or weedy species without considering the existing and desired plant community, or what effects may arise following removal of the invasive plant. Although removal of the target species is part of the restoration process, it is by itself inadequate in many situations, especially when dealing with extensive populations. Rather than implementing temporary management solutions, land managers should look to understand and alter the ecological mechanisms that are at the heart of the invasion in order to achieve desired conditions (Sheley & Krueger-Mangold 2003).

Numerous factors (e.g. climate, soil, existing plant community, and hydrology) interact following the introduction of non-native plant species that determine whether or not the species will be able to survive, grow, and reproduce in the new environment (Richardson 2004). Healthy plant communities contain a diversity of native species that occupy the majority of available spatial and temporal niches (Tilman 1986). Invasive species often become established in disturbed environments, and their presence can further increase disturbance by altering the structure and function of an ecosystem. Given a diverse native plant community, most introduced species are unable to compete for resources and survive the transition to a new locale. Certain invasive species, however, possess traits (e.g. shade-tolerance, allelopathy) that allow them to spread into intact habitats regardless of disturbance; examples include Japanese stiltgrass (*Microstegium vimineum*) and common reed (*Phragmites australis*). These aggressive invaders require an equally aggressive management and restoration program to prevent their spread and persistence in the landscape.

Restoration of native plant communities is an integral component of a comprehensive invasive species prevention and control program (NISC 2001). After removal of the target invasive species, fast-growing native species must be established to discourage growth of remaining or newly introduced invasive propagules and to stabilize the soil (Miller 2003). If the target species can be controlled with minimal disturbance to the landscape and before the ecosystem structure and processes are significantly altered, restoration can be achieved relatively quickly. However, the presence of invasive species, often coupled with human disturbance, can have multiple impacts on an ecosystem, decreasing biodiversity and altering processes associated with hydrology, soil chemistry, nutrient cycling, and sunlight availability. In these situations, restoration may need to include detailed site assessments to gather information on the inter-relationships of biotic and abiotic factors within the plant community. Depending on the scale, duration, and frequency of invasion, it may not be technically or financially feasible to restore a given area to its pre-invasion condition (NISC 2001), but managers should strive to return as much of the native plant community and ecosystem function to the restoration site as possible.

Native Plants

A native plant is one that naturally occurs in a particular habitat, ecosystem, or region without direct or indirect human influence. Native plants are crucial to national and global biodiversity conservation efforts, and their communities support healthy, productive ecosystem processes that benefit all wildlife and organisms. Additionally, native plants provide immeasurable direct and indirect benefits to the economy and society (PCA, Federal Native Plant Conservation MOU).

The Department of Defense has signed and entered into the Federal Native Plant Conservation Memorandum of Understanding (MOU) with numerous other federal agencies. The MOU established a Federal Native Plant Conservation Committee, which works to identify and recommend priority conservation needs for native plants and their habitats, and coordinates implementation of a national native plant conservation program to address those needs. The actions of the committee are guided by the following vision:

Winged sumac (*Rhus copallinum*). Photo by Adam Gundlach, Wildlife Habitat Council.

> *"For the enduring benefit of the Nation, its ecosystems, and its people; to conserve and protect our native plant heritage by ensuring that, to the greatest extent feasible, native plant species and communities are maintained, enhanced, restored, or established on public lands, and that such activities are promoted on private lands."*

Native plant communities face many obstacles related to habitat loss and degradation, competition from invasive species, and overexploitation by humans. Plants comprise greater than half of all threatened and endangered species in the United States, and Federal lands provide habitat for more than 200 listed plant species. Conservation and management of these lands is essential to preserving native plant biodiversity and to preventing additional species of plants and animals from becoming critically imperiled. Native plant conservation efforts on other public and private lands are equally important, and Federal land managers must seek innovative partnerships and outreach opportunities with public and private sector entities to conserve native plants.

The full text of the Federal Native Plant Conservation MOU is available on the Plant Conservation Alliance website.

Native Plant Resources

When acquiring native plants for restoration projects, efforts should be made to obtain plants from local nurseries, as regional variation in genotype can affect the ability of certain species to establish and persist in regions other than that which they are adapted. It is also important to plant the "true" species of each plant rather than cultivated varieties that have been breed for specific traits, such as flower color or size. Pure species strains will provide greater value to wildlife and the ecosystem.

Together, the Coevolution Institute and the North American Pollinator Protection Campaign (NAPPC) have produced factsheets regarding herbs, vines, shrubs, and trees native to the CBW, as well as the impact of invasive species on pollinators. The factsheets can be downloaded as PDFs from the Pollinator Partnership website. To locate supplies, the Lady Bird Johnson Native Plant Information Network National Suppliers Directory is a good resource.

Restoration Resources

If the native soil seed bank remains intact, controlling competition from invasive plants may be all that is needed to promote germination of native seeds lying dormant in the soil. In such cases, the treatment areas may not require replanting with native vegetation. Where feasible, germination tests can be performed on soil samples to determine whether a native soil seed bank still exists prior to initiating restoration.

In cases where the native seed bank is no longer adequate and site conditions have been significantly altered, more intensive restoration will be needed. Native plants will likely need to be reintroduced into the management area. The plant species and method of establishment to use in restoration efforts will vary depending on the region, habitat type, and site-specific features of the restoration area. A variety of organizations and resources are available to assist land managers in making sound decisions regarding specific restoration sites. The Directory of Restoration Expertise, a joint project of the Society for Ecological Restoration International (SER) and the Plant Conservation Alliance, is an integrated and comprehensive database of individuals, organizations, agencies, and companies that can provide expertise to restoration efforts. The database can be searched by a variety of parameters, including geographic area, type of restoration activity, ecosystem, type of organization being sought, and more. Similarly, the Global Restoration Network, a project of SER, hosts the Center for Invasive Plant Management (CIPM) Restoration Database. The CIPM Restoration Database provides a searchable archive of books, scientific literature, publications, on-the-ground case studies, and other documents related to invasive plant management.

The National Biological Information Infrastructure (NBII) also hosts a variety of information and resources on its Invasive Species Information Node (ISIN) covering all aspects of invasive species monitoring, identification, control, and native plant restoration.

Monitoring

An initial inventory of the management area will provide baseline data on physical site conditions, invasive species presence, degree of infestation, and plant community associations. As invasive species removal progresses and the desired plant community has been restored, monitoring is a critical post-treatment component to assess management effects and to note any changes in native and invasive plant populations (Ludke et al. 2002). Specific monitoring goals should be set (i.e. observing native plant diversity or invasive species extent). A monitoring format that produces standardized information regardless of who performs the surveys and information that relates to the restoration objectives will prove useful for long-term analysis of project success.

Regular repeated monitoring will allow new introductions of invasive species to be discovered quickly so rapid response and treatment may take place before the population becomes well established. Monitoring will also allow managers to note desirable native species that fail to establish in the restoration area. A prototype for the National Framework for Early Detection, Rapid Assessment, and Rapid Response to Invasive Species is available on the National Biological Information Infrastructure (NBII) website.

Vegetation Mapping

For installations with Geographic Information System (GIS) mapping capabilities, developing detailed maps of the restoration area will prove useful for evaluating the success of restoration efforts in future years. Maps can outline patches of different plant communities, invasive plant infestations, or land use types and can also be used to identify prime sources for invasion, such as habitat edges and corridors connecting the restoration area to adjacent areas of infestation. Stream banks, roadways, and utility rights-of-way are all potential corridors for invasive species dispersal. If surrounding lands contain invasive species, there will be constant risk of dispersal into the restoration area. Habitat patches with breaks in the natural plant cover and those adjacent to patches containing invasive species will pose the greatest risk for invasion (Reed 2004).

Once base maps highlighting plant communities and invasive species patches have been produced, other map layers showing soils, hydrology, or topography, as well as potential pathways of introduction can be overlaid. Correlations may then be drawn between invasive species population locations and landscape features.

If GIS maps are not an option, maps of the restoration site can be drawn on aerial photographs, approximating the size and location of infestations and other landscape features. Although crude in detail compared to GIS maps, the hand-drawn variety will still prove useful to future evaluations of management success.

A potentially useful resource to turn to is the Federal Geographic Data Committee (FGDC), a 19 member inter-agency committee, which coordinates development, use, sharing, and dissemination of geospatial data throughout the nation, and sets standards for metadata and mapping resources. The FGDC plans to establish the National Spatial Data Infrastructure (NSDI), which will serve as a clearinghouse to promote sharing of geospatial data between all levels of government, private and non-profit sectors, and academic institutions.

Section III: Forming Cooperative Partnerships to Achieve Management Goals

Three entities provide coordination among Federal agencies regarding invasive species issues; the Aquatic Nuisance Species Task Force (ANSTF) coordinates activities related to aquatic invasive species; the Federal Interagency Committee on the Management of Noxious and Exotic Weeds (FICMNEW) coordinates management efforts on Federal lands and provides information and recommendations regarding research, technology, and management; and the National Science and Technology Council's (NSTC) Committee on Environment and Natural Resources (CENR) coordinates research efforts (NISC 2001). Forming partnerships with the appropriate organizations can reduce the burden placed on DOD personnel and aid in accomplishing required environmental management goals.

According to Trauger et al. (1995), a partnership is a voluntary collaboration of individuals, organizations, or both to achieve common goals on a specific project within a definite time. Conservation partnerships have been used for natural resource management in watersheds throughout the country for more than 60 years (Toupal & Johnson 1998). During this period, a majority of accomplishments in the field of conservation resulted from collaborative efforts between public agencies, private organizations, and dedicated individuals (Trauger et al. 1995).

Given the mission objectives assigned to each military branch, it is often difficult to acquire funding, allocate adequate labor, and schedule time to implement invasive species control measures. The resources available to each installation and the priority for invasive species management vary depending on the size of the installation and the type of operations that they support. Management efforts undertaken by military installations to prevent, detect, and control invasive species increases the burden on personnel and diverts resources from other initiatives. Budget limitations can result in a "do more with less" approach, which often results in less being accomplished and management decisions based solely on budget considerations rather than what is most appropriate for the situation. Another common side effect of such an approach is that monitoring and follow-up treatments, which are necessary to maintain initial control levels, are left out of management plans (DiTommaso & Johnson 2006). Fortunately, many organizations exist on the local, regional, and national level to provide assistance with invasive species management. Entering into cooperative ventures can provide military installations with volunteer labor, additional sources of funding, and technical guidance in management efforts (Westbrook et al. 2005).

Though government plays a central role in partnership development (Michaels et al. 1999), developing a comprehensive approach to invasive species management requires a broad range of partnerships beyond DOD and other federal agencies. A good partnership is founded on a solid conservation need, and presently, the need to manage invasive species ranks near the top of the list. Though partnerships are not the solution in every situation, they can facilitate project completion and the advancement of programs (Trauger et al. 1995). Partnerships can also help to guarantee that invasive species monitoring and management activities are carried out on a regular basis for successive years regardless of fluctuations in available labor and funding within DOD. Long-term management is a necessary aspect of any successful invasive vegetation control program. Appendix III lists potential partnering organizations located throughout the Delaware River Basin.

Building Capacity and Incorporating the Public in Management

As outlined in Section 6j of Army Policy Guidance DAIM-ED-N (200-3) – Management and Control of Invasive Species (June 2001), installations are encouraged to enter into partnerships with other federal, state, and local agencies, tribes, and non-governmental organizations to share information, address invasive species issues, and provide public education on invasive species management to achieve local goals for controlling invasive species on and off the installation. Management of invasive species cannot be successful unless all affected landowners in a region cooperate and coordinate management actions (NISC 2001). To be effective, control efforts must involve participation from an informed public. Partnerships provide an opportunity to include local communities in complex problems that transcend political boundaries, involve multiple agencies, and require detailed knowledge of local conditions (Kenny 1999 in Leach et al. 2001).

Installation residents and local community members can be an important source of volunteer labor for management activities. In 2004, volunteers at Fort McCoy, Wisconsin contributed 1,900 hours of manual removal to invasive species control efforts, saving the base nearly $50,000. According to Kim Mello, Wildlife Biologist at Fort McCoy, outside involvement is vital to inform and educate the public about issues related to invasive species and the damage they cause.

Volunteers assisting with garlic mustard removal. Photo by Connie Gray, GA-EPPC, Bugwood.org

Invasive species are wide-ranging and do not recognize political and ecological boundaries. A cooperative invasive species program can effectively reduce invasions on military installations and surrounding lands, which in turn will help to prevent reintroduction of target species. Fort McCoy accomplishes this type of cooperative program by collaborating with the state Department of Transportation (DOT) to control spotted knapweed, synchronizing treatments on the base with treatments made to rights-of-way along local roads by DOT. Additionally, Fort McCoy holds educational outreach programs on invasive species. In collaboration with the local invasive plant working group, which Mello helped found, workshops are held at the base to train local teachers how to incorporate invasive species education into their curriculum (Westbrook et al. 2005).

In her discussion of public conservation literacy in the Journal of Conservation Biology (2001), Carol Brewer states that we cannot wait for scientific discoveries to "trickle down" to the public through textbooks and other resources. This is true of invasive species management as well, whether it is at military installations or municipal parks. Emphasis needs to be placed on creating an "environmentally literate public" (Berkowitz et al. 1997) in order to stimulate involvement in conservation initiatives. Creating opportunities for public participation in local environmental activities is crucial to promoting awareness and action on conservation issues.

In order to successfully harness the resources available in public and private entities, partnerships must be structured within a place-based framework, and they should have measurable goals. People participate in conservation partnerships because they care about 'place' – comprised of the physical location and the social, economic, and political relationships unique to each locale – and have a vested interest in the future of the place where they live. Understanding the importance of place can lead to more effective environmental partnerships. It follows that conservation partnerships must convey how they will benefit the local community and how the work contributes to the larger picture across various regional scales. In the context of this publication, military land managers in the Mid-Atlantic can describe the work being done on their installations in terms of how it contributes to the overall goals of the Chesapeake Bay Program or other smaller scale, local initiatives.

The results of a volunteer day targeting garlic mustard. Photo by Chris Evans, River to River CWMA, Bugwood.org.

Regardless of the size, each conservation partnership that focuses on invasive plant control acts like a puzzle piece on the landscape. As more pieces are added, the complete watershed restoration picture begins to form. Unfortunately, until the restoration picture is complete, each missing piece (i.e. unmanaged infestation) has the potential to reinfest areas already under management or not yet invaded. This is why it is imperative to increase public awareness and involvement in conservation activities so management can take place across the largest area possible.

Where to Turn for Support

Conservation organizations on both the national and local levels are often valuable resources for invasive species management. Federal and state government agencies can provide expertise and equipment, and may be able to provide funding for certain types of projects. Colleges and universities are typically at the cutting edge of invasive species research and can provide astute recommendations for management. University faculty may also represent potential partnership opportunities, as military installations could act as sites for research studies.

Volunteer involvement is crucial in the fight against invasive plants and can offer significant benefits to control programs, including reduced dependence on chemicals. Installation residents and residents of surrounding communities offer the most convenient source of volunteers for management programs. Volunteer outings can be coordinated with school groups, scouts, or similar youth groups, where education can be integrated into management activities. If installation residents are not an option, local conservation groups are often open to new opportunities and may already coordinate volunteer work days in the area.

When using volunteer labor for management, it is important to train volunteers to properly identify the target species, avoid desirable species, and limit soil disturbance to the greatest extent possible. Sending a group of untrained volunteers onto the landscape may result in more harm then good. It is also important to be flexible with volunteers. It is best to schedule work days at times that are convenient for the group. Although management may be focused in a particular area, try to shift locations on occasion to prevent the work from becoming monotonous. To keep morale high, it also helps to incorporate an activity, even if just for a few minutes at the end of the event, that gives volunteers a tangible experience of restoring habitat. Even with the understanding the invasive species removal is critical for habitat restoration and preservation, it's easy to feel defeated in the seemingly endless battle against invasives. It helps to do a native planting (even when not strictly necessary for native plant community regeneration) or other habitat enhancement activity after removing invasives. For an inexpensive habitat enhancement activity, you can use woody debris from invasive species removal to build brush piles or create basking logs. (Important note: some species, such as tree of heaven, can resprout from woody debris -- most species won't, but to be safe, don't ever move to debris to a new area, and always monitor for resprouts!)

The U.S. Fish and Wildlife Service recently developed a website to provide guidance on incorporating volunteers into management efforts. The website is divided into several informational modules with topics ranging from the invasion process to tips on how to effectively present information to volunteers.

Although management must take place consistently throughout the year, there are many annual events that provide great opportunities for volunteer involvement. Installations can host community outreach events during National Invasive Weed Awareness Week (NIWAW), which is held each year during February or March. Numerous presentations and activities are held in Washington, D.C. during NIWAW, but local events held around the country are critical to magnifying the impact of the awareness week. For installations with natural areas that are open to public access, invasive species management events can be held on National Public Lands Day (NPLD). This annual event, which began in 1994, educates the public about pressing environmental issues and builds partnerships to enhance and restore public lands. Each year, tens of thousands of volunteers participate in NPLD across the country. Earth Day is another well known day of action that is an opportune time to hold a volunteer work day.

Locating Potential Partners

The Mid-Atlantic Exotic Pest Plant Council website contains an extensive directory of links to conservation-related organizations in the region. In general, excellent places to begin looking for partners are: county Master Gardener / Master Naturalist programs, local schools, scout troops, native plant societies, and invasive species groups such as cooperative weed management areas.

Expertise within DOD

Although collaboration between other government agencies, private entities, and the general public is important, do not overlook the wealth of experience, information, and expertise available within the military. Many installations are dealing with similar environmental management concerns and objectives, and are familiar with the regulations and restrictions intrinsic to DOD, making them valuable resources to consult with regarding management issues. In times when expanded mandates and reduced budgets force natural resource managers to find new ways of accomplishing goals (Trauger et al. 1995), partnering with other DOD installations to identify efficient and effective approaches to management objectives may be a vital link in the conservation chain. Joint military efforts may also present opportunities to share personnel, equipment, and technology on management initiatives.

For guidance on invasive species management, refer to the:

- Armed Forces Pest Management Board (AFPMB)
- U.S. Army Corps of Engineers (USACE), Engineer Research and Development Center (ERDC)
- U.S. Army Environmental Command (USAEC)
- Center for Environmental Management of Military Lands

Section IV: Case Studies

In conjunction with this publication, on-the-ground control of invasive plant species was initiated at three pilot sites at DOD installations. Due to the office location of the Wildlife Habitat Council (whose staff did on-the-ground work in conjunction with military personnel and community partners at the pilot sites), the sites were located in the Chesapeake Bay Watershed rather than the Delaware River Basin. Still, the outcomes of the pilot projects can be instructive for land managers in the basin with regard to management planning strategies, program implementation and volunteer coordination. Following is a description of each pilot site, management methods used, and lessons learned.

I. Fort Belvoir Army Garrison, Virginia

Fort Belvoir is located approximately 20 miles south of Washington, D.C. along the Potomac River. The installation comprises more than 8,500 acres, and an invasive species survey conducted by Paciulli, Simmons and Associates (2006) shows a wide variety of invasive plants found throughout Fort Belvoir, with varying degrees of infestation and management urgency.

The program at Fort Belvoir was the longest-running of the three pilot programs, because it was initiated in 2006 as part of Legacy project 06-328. The Fort Belvoir model was replicated at NSF Indian Head and NSF Carderock in 2008-2009 as part of Legacy project 08-328 (which also included continuation of the Fort Belvoir program). The programs at NSF Indian Head and NSF Carderock benefitted greatly from the lessons learned at Fort Belvoir, as detailed below.

Site description

The site on Fort Belvoir chosen for the invasive species management pilot program stretches along the basin trail in the Accotink Wildlife Refuge (AWR). The AWR is open to public access and has more than 8 miles of interconnected recreational trails that wind through the varied habitats of the AWR. The portion of the basin trail that traverses the pilot site is situated adjacent to Accotink Bay, which empties to the Potomac River via Gunston Cove. The area of focus for the pilot site is approximately 15 acres in size and contains open meadows, floodplain forest, and upland forest dominated by mature American beech (*Fagus grandifolia*).

The pilot site contains several invasive plants identified in the Paciulli, Simmons and Associates report as being in the establishment phase of invasion and of moderate to high management urgency. Chinese lespedeza (*Lespedeza cuneata*), a perennial semi-woody herb, has invaded meadow areas near the trailhead and currently forms a large portion of the vegetative cover in these areas. Japanese honeysuckle (*Lonicera japonica*), a perennial invasive vine, is ubiquitous throughout the pilot site and can remain green year-round given mild winter temperatures. In certain areas it forms a nearly monotypic groundcover in the forest understory and often can be observed climbing trees into the canopy. Japanese stiltgrass (*Microstegium vimineum*), a persistent and pernicious annual grass species, is common along the basin trail and in the surrounding forest understory, forming monotypic patches to the exclusion of native species. Oriental (or Asiatic) bittersweet (*Celastrus orbiculatus*), which is another perennial invasive vine, has scattered patches along the trail, and readily climbs trees into the canopy. Other invasive species, including common reed (*Phragmites australis*), English ivy (*Hedera helix*), and mimosa (*Albizia julibrissin*), had a minor presence and are likely only in the dispersal phase of invasion at the pilot site.

Management Areas and Treatments

Several areas within the pilot site were the focus of control work; they included a significant infestation of mixed Japanese honeysuckle and stiltgrass covering more than an acre, patches of Japanese stiltgrass lining the trail and its near vicinity, scattered patches of oriental bittersweet as well as a larger patch of bittersweet disjunct from the main pilot site, the lespedeza-infested meadows, and specimens of English ivy and mimosa wherever they occurred throughout the pilot site. Manual removal of plants comprised much of the control work, as Fort Belvoir only allows state-certified applicators to perform herbicide applications. Manual removal (hand pulling) was used for Japanese stiltgrass patches along the trail, a demonstration plot carved out of a thick groundcover of Japanese honeysuckle, two small patches of English ivy, and mimosa seedlings throughout the site. Later in the project, Invasive Plant Control, Inc. was contracted to perform herbicide applications on larger areas of Japanese honeysuckle, Japanese stiltgrass, and oriental bittersweet.

Japanese stiltgrass/honeysuckle infestation: One of the worst areas of infestation at the pilot is an area of lowland forest between the trail and Accotink Bay infested by Japanese honeysuckle and Japanese stiltgrass. When first viewed in late summer of 2006, the area was dominated by these two species, with few other species growing in the area. Young honeysuckle vines could be seen overtopping low-growing vegetation and mature vines climbed well into the canopy. Drought-like conditions in 2007 reduced the vigor of the stiltgrass and honeysuckle; however, they remained the primary vegetative cover through the growing season.

Mature honeysuckle vines extend into the canopy of a host tree. Photo by Adam Gundlach, Wildlife Habitat Council.

In the spring of 2007, manual removal of Japanese honeysuckle began in a small demonstration plot covering approximately 200 square feet. The honeysuckle existed mostly as a groundcover,

Manual removal demonstration plot (foreground) with infestation in background. Photo by Adam Gundlach, Wildlife Habitat Council.

but any vines found growing up trees were cut. As vines were pulled up from under the leaf litter, as much of the root and rhizome structure was removed as possible and desirable vegetation was avoided. The area was then monitored throughout the growing season to remove any remaining root fragments that resprouted. In the fall of 2007, a crew from Invasive Plant Control, Inc. (IPC) performed a foliar herbicide application across much of the area using a glyphosate-based herbicide (Razor® Pro) plus adjuvants.

Efficacy of the control measures was assessed in 2008. The cutting of vines appeared to have been beneficial; the trees in the demonstration plot were in good health and few woody vines had reestablished on the trunks. However, honeysuckle sprouts had retaken most of the understory area. Some native forbs were sighted growing amongst the honeysuckle, suggesting that the clearing of the honeysuckle cover had been beneficial in allowing for germination of native wildflowers. Despite the encouraging rebound of native forbs, manual removal of honeysuckle in the understory was determined to be too time-intensive compared to gains made to be continued. Unfortunately, due to installation procedures and the difficulty of communicating with personnel (see "Obstacles Encountered and Lessons for Future Projects" below), continued herbicide treatment was not an option either. This plot was thus abandoned in 2008 – other than cutting of the few remaining woody vines growing up tree trunks – in favor of concentrating on sites with higher potential for achieving control with available resources.

Japanese stiltgrass patches: Japanese stiltgrass grows in dense patches along the trail and in the surrounding forest understory. Due to its shallow, poorly developed root system, stiltgrass is easily pulled by hand, and manual removal was the main control method employed. Pulling was initiated in 2007 and repeated in 2008. Several volunteers from the Friends of Accotink Creek, a local non-profit volunteer organization, assisted with stiltgrass removal. Patches located on or immediately adjacent to the trail were of the highest priority in order to prevent seed production and possible dispersal by trail users. In summer prior to seed set, stiltgrass was pulled and piled together near the removal area. In early fall, as the stiltgrass began to flower and produce seed, all plants pulled were bagged and removed from the site. As the priority patches were removed, work shifted to areas farther from the trail, including an area along a small woodland stream, which also could act as a dispersal route for seeds.

The photo on the left was taken prior to removal (right) of Japanese stiltgrass along the basin trail. Photos by Adam Gundlach, Wildlife Habitat Council.

Although Japanese stiltgrass is an annual species and removal efforts prevented seed production along much of the trail for two growing seasons, the seed bank is known to persist for up to five years, so repeated efforts in future seasons will be necessary. Future control efforts may integrate a pre-emergent herbicide application to prevent germination of stiltgrass seeds, which may allow native species an opportunity to become established.

Oriental bittersweet: Several small areas of oriental bittersweet were located along the trail. Though none of the patches were large in extent, most had already climbed well into the canopy and would likely engulf entire trees within a few years. In 2007, a crew from Invasive Plant Control, Inc. treated these bittersweet patches by cutting vines near ground level and immediately treating the stumps with a 25 percent mixture of Garlon 3A to kill both above and below ground portions of the plants.


Oriental bittersweet infestation climbing into the canopy. Photo by Adam Gundlach, Wildlife Habitat Council.

An extensive oriental bittersweet infestation located a short distance from the pilot site was also slated for removal. Control work began on this area in the fall of 2007, but was not completed due to poor weather conditions and time shortage.

In 2008, monitoring and follow-up control took place. Remaining living woody vines were cut at ground level to free the native trees and shrubs underneath. Berries were collected and disposed to prevent birds from spreading the seed.


English ivy creeping growth along the ground and up the trunk of a pine tree in the background. Photo by Adam Gundlach, Wildlife Habitat Council.

English ivy: Two small patches of English ivy were identified during initial survey of the pilot site in 2006. Most vines were spreading through the leaf litter and were pulled by hand, removing as much of the stem and root structure as possible. All vegetative material was bagged and removed from the site to prevent sprouting from stem fragments. Follow-up monitoring was conducted in subsequent seasons to look for regeneration.

Lespedeza-infested meadows: The meadows are located immediately beyond the trailhead at the entrance to Accotink Wildlife Refuge. They border Accotink Bay, which boasts a myriad of avifauna during the breeding season. The meadows were originally intended to be a main focus of control efforts. According to Fort Belvoir


Meadow infested by Chinese lespedeza. Photo by Adam Gundlach, Wildlife Habitat Council.

personnel, they have exhibited significant decline in wildflower diversity and abundance in recent years. This decline correlates directly with the introduction and spread of Chinese lespedeza, a species that has become common in many open habitats at Fort Belvoir.

One of the meadows was scheduled for an herbicide application in late summer of 2007; however, delays in the approval process forced the treatment to be cancelled, as the optimal application time passed and the lespedeza began to produce seed and senesce. In 2008, delays in approvals were again an issue, but ultimately a degree of success was achieved in having the installation's grounds crew mow the meadows prior to seed set. Ideally, mowing would take place in the flower bud stage. The 2008 mowing was slightly late but should be a good start in weakening the infestation and will contribute to reducing the lespedeza's vigor and dominance if mowing is performed at the bud stage in subsequent years. Installation personnel are aware of the necessity of repeated mowing and plan to facilitate it in future years.

Obstacles Encountered and Lessons for Future Projects

As with any project, work at the pilot site encountered its share of bumps in the road as well as positive outcomes. Lessons learned from the project are detailed below.

Make Sure Priorities Mesh: Installation approval processes and communications with personnel were serious obstacles in this project. Fort Belvoir is currently undergoing BRAC, and it may have been wise to consider the implications of this process before investing resources in a program at the installation. Throughout the term of the project, the natural resources manager was necessarily consumed by BRAC and voluntary programs such as this one were low priority. This is not to say personnel were not helpful and interested when they did have time to devote – for example, one of the contractors facilitated the mowing of the lespedeza meadows -- but the amount of work the staff had and the installation's lengthy approval processes made it difficult to move the invasive species management program forward. Proposed volunteer events and workshops were delayed, cancelled or denied approval. Control actions were cancelled due to failure to receive approval before the window of time when treatment would be effective passed.

The lesson learned from this experience is that it is crucial to make sure all partners are fully able to commit to the program before it begins. It may be helpful to outline, in writing, the responsibilities of each partner before work begins. That way, it would be possible to foresee potential roadblocks and allocate resources to projects that further everyone's goals and have greatest support from all parties.

Plan Herbicide Treatments Well in Advance: The project was initiated in mid-July of 2006 and Fort Belvoir was not confirmed as the location for the pilot site until early September of the same year. The late start in 2006 prevented treatments from taking place during that growing season, as the optimal time to treat most species had passed. In 2007, delays in approval for herbicide applications and weather conditions prevented certain target species from being treated, further reducing potential results that could have been achieved. The treatments that were implemented during 2007 showed promising results, but repeating them in 2008 proved difficult due to issues communicating with installation personnel.

These experiences highlight the importance of planning treatments well in advance. It can take many months for paperwork and approvals to be completed, and once approvals are in place, it takes time to get an appointment with herbicide contractors, and there must be a cushion of time built in to the schedule to accommodate unpredictable weather conditions. It is best to maintain regular contact with all parties involved in management activities to ensure that everyone is on the same page and all logistical details are addressed.

Work with Available Resources: Funding for invasive species management may often be at a premium, but progress can still be made toward management goals. Although a lack of funding may prevent large-scale invasive species control, small-scale efforts can be accomplished in the interim until funding is available. As described in the previous section, volunteers can be a vital resource in control efforts, and their work can reduce reliance on chemical treatments. Manual removal performed by volunteers will not be suitable in every situation, especially for extensive infestations, but the alternative of taking no action may be even more detrimental. The effectiveness and benefit of persistent small-scale controls should not be overlooked – such efforts carried out over several years can obtain significant results. Being flexible and utilizing what resources are available will provide greater opportunities for management success.

II. Naval Support Facility Indian Head, Maryland

NSF Indian Head is located in Charles County, MD, approximately 30 miles south of Washington, DC. The installation occupies approximately 3,500 acres on the eastern bank of the Potomac River. It is comprised of a main parcel of about 2,000 acres, an annex (known as Stump Neck) of about 1,000 acres separated from the main parcel by Mattawoman Creek, and three other small parcels nearby.

The program at NSF Indian Head was the second pilot program to be initiated. The installation was chosen as the location for the second pilot program due to its enthusiastic staff and abundance of natural areas that could be valuable for both installation operations and wildlife habitat if invasive species infestations are controlled. The program began in spring 2008.

Site description

Following a survey of potential project areas identified by the installation's natural resources personnel, a 3.77-acre seasonal wetland at the Stump Neck annex was identified as the optimal project location. The area has a diversity of herbaceous and woody vegetation and provides habitat for a wide variety of species, including bald eagles (*Haliaeetus leucocephalus*). Forested areas surround the wetland, adding habitat diversity to the already important wildlife area. In fact, the Stump Neck forests are known to provide breeding habitat for 16 of the 21 forest interior dwelling bird species in southern Maryland (DOD Partners in Flight 2009). However, the wetland is host to a serious infestation of Japanese stiltgrass that has taken over large portions of the wetland and is beginning to spreading into the adjacent forested areas. Due to the opportunity to not only restore native plant communities in the wetland but also protect the forest from invasion, this site was chosen as the location for the pilot program.

An additional reason for initiating the program in this particular area was that the wetland spans the border with neighboring Smallwood State Park, and the ranger who manages the habitat on the park side expressed interest in partnership. The site thus presented an opportunity for collaborative management, which would allow for greater impact – due to a potential additional source of manpower and resources – as well as reduced chance of reinvasion from the adjacent property and the creation of a high-quality cross-property corridor for wildlife to move.

Spraying a monoculture of stiltgrass (adjacent to a patch of ground ivy, top left corner) at NSF Indian Head. Photo by Susan Reines Robinson, Wildlife Habitat Council

Management Areas and Treatments

Japanese stiltgrass: Hand-pulling in areas where stiltgrass was growing amongst natives commenced in mid July. A crew from Invasive Plant Control, Inc. came to spray monocultures (Accord 1%) in August. Follow-up hand pulling continued through September until the seed heads opened (for specimens removed after seed heads appeared but before they opened, seed heads were bagged and discarded). An intern from Salisbury College helped with the hand pulling. A weed-whacker was also employed to remove monocultures not controlled during the herbicide treatment. In November, a crew from the Maryland Conservation Corps came out to help plant native species with the Wildlife Habitat Council staff and NSF Indian Head personnel.

A volunteer from the Maryland Conservation Corps and the NSF Indian Head natural resources manager (background) plant native species amongst the stiltgrass debris left after the herbicide treatment. Photo by Susan Reines Robinson, Wildlife Habitat Council.

While the hand pulling and herbicide treatment were effective in removing stiltgrass, it was clear when the seed heads emerged in fall that the methods had not removed all stiltgrass plants and some seed would be dispersed. Control was most effective in small patches in the forest, at the invasion's expanding front, where it was possible to remove all plants in a given area.

Ground ivy: Ground ivy (*Glechoma hederacea*) was present throughout the wetland in open, seasonally dry areas. It was sprayed during the herbicide treatment when found near stiltgrass. Hand removal was considered during the winter but was ultimately judged to be an inefficient use of resources (time) due to the large area throughout which the species was spread and its relatively minimal disturbance to the habitat compared to the other invasive species at the site.

Japanese honeysuckle: Japanese honeysuckle (*Lonicera japonica*) was overtopping many of the native shrubs in the wetland, including spicebush (*Lindera benzoin*) and highbush blueberry (*Vaccinium corymbosum*), two important species for birds and other wildlife. Vines that were strangling native trees or shrubs were cut near the ground in winter, when they were easily visible and accessible. The extent to which these vines will regenerate and move up the shrubs/trees again is not yet known, but regeneration and the need for continued cutting is expected due to the fact that applicator certification requirements prevented the application of herbicide to the cut vines.

Obstacles Encountered and Lessons for Future Projects

Delineate a small project area: After spending the summer and fall working at the 3.77-acre wetland, it became clear that, given available resources, program impact may have been greater if resources had been concentrated in a smaller area or on a less severe invasion. The project did benefit native wildlife in the wetland by clearing space for native plants to spread and by reducing stiltgrass seed dispersed that year. Yet some stiltgrass plants did go to seed, despite spending about half of the money budgeted for all three sites' herbicide treatments and performing manual controls almost weekly. The most effective results were achieved at the expanding front of the invasion, in the forest, where entire patches could be removed as they emerged to prevent the grass from moving further into the forest. In retrospect, it may have been more effective to concentrate all resources on the invasion front rather than tackling the wetland directly. On the other hand, if focus had been solely on the forest, no enhancements to the native plant community in the wetland itself would have been achieved, and that would have been a shame due to the wetland's importance to wildlife and the health of the watershed as a whole.

Another reason it might have been better to concentrate on a smaller area is that volunteers who helped at this site often expressed frustration at the overwhelming invasive population. Morale probably may have been better, and more volunteers may have returned to participate in follow-up events, if they had been working in a smaller area where progress was visible.

Have multi-year resources secured before implementing controls: Stiltgrass has a seed bank that persists up to five years. The funding for project 08-328 was limited and it was known that it would last for only one growing season, an insufficient amount of time to control any invasive species. In the case of NSF Indian Head, controls were implemented anyway because the installation's natural resources personnel were on board with the project and could monitor and maintain the area in future years, and there was strong potential for collaborative management with the park, which could add further resources. Unfortunately, things can happen – such as the global economic crisis, which has hit DOD natural resources budgets hard. The natural resources personnel may yet find a way to carry on with the project, but this year everyone is trying to do more with less. The park has not yet been able to contribute any resources except facilitation of participation by the Maryland Conservation Corps.

Keeping the management area smaller, as discussed above, would have helped alleviate this problem by making it easier for installation personnel to perform follow-up controls on a tight schedule. Funders could also help to allay this issue by funding invasive species programs for multiple years, thereby ensuring return on the first year's investment.

III. Naval Support Facility Carderock, Maryland

NSF Carderock is in Bethesda, MD, in the northwest suburbs of Washington, DC. Although Bethesda is a developed suburban area, and although the installation covers less than 200 acres, NSF Carderock is a key property for wildlife habitat because the surrounding land uses include two parks and forested buffer area owned by the county. With natural areas on almost every side, the installation can be an integral part of a larger wildlife corridor.

NSF Carderock was the third pilot program implemented, but its results are arguably most successful, as it was set up using lessons learned at the other two sites. Work began in summer 2008 and was in full swing by fall 2008.

Site description

Prior to implementation of this program, the NSF Carderock was already partnering with The Nature Conservancy to control Bradford pear trees. The trees on the installation were suspected of serving as a seed source for the invasive pears to colonize nearby locations in the Potomac Gorge, home to one of the highest concentrations of globally rare natural communities in the nation. As The Nature Conservancy was charged with protecting the gorge, its staff was working with NSF Carderock personnel to eliminate the seed source on the base.

With that priority area already under management, the NSF Carderock natural resources manager suggested that the Wildlife Habitat Council program be implemented at the installation's constructed wetland. The wetland is situated in the installation's northeast corner. The area is comprised of a seasonally wet meadow, a permanent pond, and a vegetative buffer that is herbaceous on the pond banks and becomes shrubby to forested as it stretches up to the installation's fenceline, where it meets the county-owned portion of the forest buffer. The wetland area was supporting a variety of native flora and fauna before this program began, but a number of aggressive invasive species were beginning to dominate large portions of the habitat. Canada thistle and crown vetch covered approximately 80% of the pond banks. A phragmites patch sat in the middle of the wet meadow. Bush honeysuckle was spreading through the shrubby portion of the buffer, and invasive vines, particularly oriental bittersweet, were overtopping trees in the forested portion.

Management Areas and Treatments

Crown vetch: A group of volunteers, including NSF Carderock employees (who came in on a Saturday to help) Montgomery County Master Gardeners, and other community members dug the crown vetch from the pond banks in October. They planted native species to stabilize the soil and add wildlife value. The Wildlife Habitat Council staff returned many times (almost weekly) over the next few months to manually remove resprouts. Only a few sprouts emerged in spring, perhaps because, unfortunately, a number of other invaders moved in to quickly take the crown vetch's place, as detailed below.

Canada thistle: Canada thistle, which had existed on the pond banks in scattered clumps of about 10 feet by 5 feet prior to the crown vetch removal, spread vigorously once the competing invader was removed. Wildlife Habitat Council staff, Master Gardeners, and other volunteers, dug it from the banks, returning weekly through early winter to remove resprouts from root segments.


Sumacs emerge amongst the dead thistle left after the herbicide treatment on the pond banks. Photo by Susan Reines Robinson, Wildlife Habitat Council.

Despite these efforts, the invasion came back worse than ever in spring 2009. At this point, chemical control was deemed necessary. A crew from Invasive Plant Control, Inc., sprayed the thistle with Garlon 3A 2% in April. This treatment ideally would have taken place slightly earlier, when the thistle was green but most natives had not leafed out, but an earlier scheduled treatment had to be postponed due to rain. Still, with the contractors being extremely careful to avoid damaging natives, the herbicide treatment was very successful. The few thistle resprouts that emerged later in the spring were dug out very carefully by hand. A few weeks after the herbicide treatment, early successional natives such as sumacs (*Rhus* spp.) emerged, as pictured at left.

Phragmites: Prior to implementation of this program, NSF Carderock personnel had been digging a patch of phragmites (approximately 20 feet by 15 feet) out of the wetland each winter. The manual removal had succeeded in keeping the patch from spreading, even if it hadn't eliminated it. Herbicide treatment was considered in 2008 but was not possible due to weather conditions, so Wildlife Habitat Council staff dug this patch by hand, being careful to remove all parts of the rhizomes. Currently, it appears that the removal was successful, but at least some regeneration is expected.

Bush honeysuckle & oriental bittersweet: Another component of this program, in addition to the on-the-ground work and production of this publication, was a seminar held at NSF Carderock in April. The seminar focused on creating conservation partnerships for watershed protection, with particular focus on invasive species management and native plant community restoration. Representatives of a variety of government agencies, community organizations, and 14 military installations attended. One outcome of the seminar was a partnership with the Montgomery County

Department of Environmental Protection. As mentioned earlier, the county owns a portion of the forest buffer adjacent to the wetland. In June, county DEP employees joined NSF Carderock personnel, staff of the Wildlife Habitat Council and The Nature Conservancy, and community volunteers to enhance the forest buffer on both sides of the installation/county fenceline. Participants cut bush honeysuckle and oriental bittersweet vines at ground level, and certified herbicide applicators from The Nature Conservancy applied herbicide to the cut stumps. Following the invasive species removal, participants planted native species that serve as host plants for pollinators and added structural components such as basking rocks to benefit butterflies. A follow-up partnership field day is planned for September.

Obstacles Encountered and Lessons for Future Projects

As detailed in the previous sections, it became clear from experiences at the first two pilot sites that invasive species management programs can easily be derailed by security obstacles, inattentive or overburdened natural resources personnel, lack of volunteer support, and overambitious delineation of project sites. For the program at NSF Carderock, there obstacles were addressed by:

1) Delineating small, manageable project areas where participants could see progress each workday. The management areas were expanded as the program progressed.
2) Locating a few key supporters within the installation and in the community to rally volunteer activity. The natural resources personnel at NSF Carderock and up the chain of command were enthusiastic, involved and accommodating. Additionally, the program benefited from taking place at an installation in a populated area where there are many conservation groups. A few supportive individuals within the Montgomery County Master Gardeners and the Montgomery County Department of Environmental Protection made a huge difference in rallying community volunteers. Hitting upon these contacts was partly luck, but it illustrates the point that making a few key connections can get the ball rolling for a successful program.
3) Concentrating activities in areas where it is easiest to get approvals for work and security access for volunteers. At Fort Belvoir, the installation is open (visitors just need to present ID at the gate), but the installation's complex approval processes for control actions and events made progress difficult. At NSF Indian Head, approval processes were not as much of an issue, but the site's location in a remote, restricted area did impact volunteer interest. At NSF Carderock, the security team was extremely helpful and approval processes went smoothly, and there are abundant community organizations, schools, and other sources of volunteers nearby.
4) Making sure volunteers understand that they are not just pulling weeds – they are restoring habitat. After obtaining feedback that volunteers at NSF Indian Head felt overwhelmed, it was made policy for the program to always incorporate another activity into an invasive species removal event. It could be as simple as a small planting at the end of the day or creation of a brush pile using debris from woody invasive plants. The active building up of habitat components was observed to have strong impact on volunteer morale.

Finally, it must be emphasized that a significant reason the NSF Carderock program went so smoothly was that there was support up the chain of command. The commander of Naval Support Activity North Potomac, Scott Merritt, strongly supported the program and helped to make sure approvals, gate access, and other potential obstacles did not interfere with the program. With the motivation and support of an officer of his level, everyone was particularly motivated to make habitat restoration happen.

Appendix I – Terrestrial Herbicides

HERBICIDE	BRAND NAMES	TARGET SPECIES	NOTES
2,4-D	Navigate®, Savage®, WeedDone®	Various broadleaf species	Systemic herbicide available in various formulations. Often combined with other herbicides.
Clopyralid	Transline®, Reclaim®, Curtail®, Stinger®	Annual and perennial broadleaf species	Highly selective alternative to picloram. Do not apply to water. Soil activity may allow it to leach into groundwater.
Fluazifop-p-butyl	Fusilade DX®, Fusion®, Tornado®	Annual and perennial grass species	Systemic, foliar herbicide. Compatible with other herbicides. Toxic to fish.
Fosamine	Krenite®	Trees and shrubs	Inhibits bud development. Defoliant used to “trim” portions of trees and brush.
Glyphosate	Roundup Pro®, Rodeo® (aquatic uses), Accord® (aquatic uses)	Wide variety of woody, broadleaf herbaceous, and grass species	Non-selective, broad-spectrum, systemic, foliar herbicide. Use appropriate formulation near water.
Hexazinone	Velpar®, Pronone®	Annual, biennial, and perennial species, as well as certain woody species	Systemic herbicide. Requires rainfall or irrigation to activate. Potential for ground water contamination from leaching. Toxic to algae.
Imazapic	Plateau®, Plateau Eco-Pak®, Cadre®, Journey®	Annual and perennial species	Soil active, providing residual control for certain species. Avoid use in root zone of desirable species. Effectiveness varies depending on plant.
Imazapyr	Arsenal®, Sahara®	Most grass, broadleaf, vine, and many shrub species	Soil active, systemic herbicide. Provides residual control of many species. May affect non-treated species through root uptake.
Picloram	Access®, Pathway®, Tordon® K	Woody and herbaceous broadleaf species	Soil active, systemic herbicide. Environmental persistence may allow leaching and surface runoff.
Sethoxydim	Poast®, Vantage®	Annual and perennial grass species	Selective herbicide. Rapid degradation in sunlight can limit effectiveness. Toxic to aquatic organisms.

Herbicide	Brand Names	Target Species	Notes
Triclopyr	Garlon®, Remedy®, Access®, Crossbow®, Pathfinder®	Woody and broadleaf species	Ester formulation is highly toxic to aquatic organisms. Can be combined with other herbicides (picloram, 2,4-D).

Compiled from Tu et al. 2001, http://www.kellysolutions.com/, http://www.cdms.net, http://extoxnet.orst.edu/

Disclaimer: Always read the entire herbicide label, including supplemental labeling materials. Follow all transport, storage, mixing, and application guidelines, and wear all recommended personal protective equipment. Be sure to follow all local, state, DOD, and installation regulations regarding herbicide application.

Appendix II – Aquatic Herbicides

Many of these products cannot be applied directly to water, but may be used to treat vegetation in or near aquatic environs under the appropriate conditions. Brand names listed do not represent all available commercial formulations. Treating large areas of aquatic weeds can result in depleted dissolved oxygen as plants decompose, and may result in fish kills.

HERBICIDE	BRAND NAMES	TARGET SPECIES	NOTES
2,4-D	Aqua-Kleen®, Weedar® 64, Navigate®	Annual, biennial, and broadleaf species, aquatic macrophytes	Selective for broadleaf species. Often used in combination with other herbicides. Available in various formulations.
Complexed Copper, chelated copper	Cutrine®-Ultra, Komeen®, K-Tea®	Algae and certain aquatic macrophytes	For use in slow moving or stationary bodies of water. Often combined with other aquatic herbicides.
Dicamba	Veteran® 720	Woody and herbaceous broad leaf species	Do not apply directly to water or to irrigation canals. Only apply to emergent vegetation. Toxic aquatic invertebrates.
Dichlobenil	Casoron®	Wide range of woody and herbaceous broadleaf and grass species, and certain aquatic species	Non-selective, soil active, systemic herbicide. Apply only in stationary waters. Even application is essential for effectiveness.
Diquat	Reward®, AQUA-TRIM™ II, Weedtrine®-D	Algae, aquatic macrophytes, and other broadleaf and grass species (depending on formulation)	Non-selective, fast-acting, contact herbicide. Apply only in stationary waters. Toxic to aquatic invertebrates.
Endothall	Aquathol® K, Aquathol® granular, Hydrothol®, Accelerate®	Algae, submergent and emergent aquatic species	Selective contact herbicide with relatively short environmental persistence.
Fluridone	Sonar®, Avast!®	Emergent and submergent aquatic species	Slow-acting, systemic herbicide. Requires long contact period, which may preclude use in flowing waters.
Glyphosate	Rodeo®, Accord®	Emergent and floating aquatic species, and all terrestrial species	Non-selective, broad-spectrum, systemic herbicide that is absorbed through foliage.
Imazapyr	Arsenal®, AquaPier®	Wide range of woody and herbaceous species	Non-selective, residual soil activity for pre-emergence control.
Tebuthiuron	Spike®	Woody species	Do not apply directly to water. Soil active; avoid application to root zone of desirable species.

HERBICIDE	BRAND NAMES	TARGET SPECIES	NOTES
Triclopyr	Renovate® OTF	Emergent, submergent, and floating aquatic species	Not to be applied to salt-water bays or estuaries, or irrigation canals.

Compiled from Aquatic Plant Information System (APIS), http://www.kellysolutions.com/, http://www.cdms.net, http://extoxnet.orst.edu/

Disclaimer: Always read the entire herbicide label, including supplemental labeling materials. Follow all transport, storage, mixing, and application guidelines, and wear all recommended personal protective equipment. Be sure to follow all local, state, DOD, and installation regulations regarding herbicide application.

Appendix III – Management Resources

INVASIVE SPECIES INFORMATION AND ORGANIZATIONS

Bugwood Network - Invasive and Exotic Insects, Diseases, and Weeds: Information and Images: http://www.invasive.org/

Catskill Regional Invasive Species Partnership (CRISP)
http://www.catskillcenter.org/programs/land/crisp.html

Catskill Streams
http://www.catskillstreams.org/stewardship_streamside_is.html

Center for Invasive Plant Management (CIPM):
http://www.weedcenter.org
CIPM Restoration Database:
http://www.globalrestorationnetwork.org/database/cipm-database/

Delaware Invasive Species Council
http://www.delawareinvasives.net/

Federal Interagency Committee for the Management of Noxious and Exotic Weeds:
http://www.fws.gov/ficmnew/

Federal Noxious Weed Disseminules of the United States:
http://www.lucidcentral.org/keys/v3/FNW/

Global Invasive Species Information Network (GISIN):
http://www.gisinetwork.org/

Inter-American Biodiversity Information Network (IABIN):
http://www.iabin-us.org/
Invasive Information Network (I3N):
http://i3n.iabin.net/

Invasive Plant Atlas of New England (IPANE):
http://nbii-nin.ciesin.columbia.edu/ipane/

Invasive Plant Control, Inc.:
http://www.invasiveplantcontrol.com/

Invasive Plant Council of New York State:
http://www.ipcnys.org/

Mid-Atlantic Exotic Pest Plant Council (MA-EPPC):
http://www.ma-eppc.org/

National Association of Exotic Pest Plant Councils:
http://www.naeppc.org/

National Biological Information Infrastructure (NBII):
http://www.nbii.gov/
  Invasive Species Information Node (ISIN):
  http://invasivespecies.nbii.gov/library.html

National Invasive Species Information Center:
http://www.invasivespeciesinfo.gov/
  State-specific Resources:
  http://www.invasivespeciesinfo.gov/unitedstates/state.shtml

The Nature Conservancy (TNC) Invasive Species Initiative:
http://www.invasive.org/gist/

Pennsylvania Department of Conservation and Natural Resources
Invasive Plants in Pennsylvania:
http://www.dcnr.state.pa.us/forestry/wildplant/invasive.aspx

Pennsylvania Integrated Pest Management Program (PA IPM):
http://paipm.cas.psu.edu/

Plant Conservation Alliance Alien Plant Working Group
Weeds Gone Wild: Alien Plant Invaders of Natural Areas:
http://www.nps.gov/plants/alien/index.htm

Smithsonian Environmental Research Center – Marine Invasions Research Lab
  National Exotic Marine and Estuarine Species Information System (NEMESIS):
  http://invasions.si.edu/nemesis/

  Aquatic Invasions Research Directory:
  http://invasions.si.edu/aird/

  Nonindigenous Species Database:
  http://invasions.si.edu/aird/

Society for Ecological Restoration International: Directory of Restoration Expertise:
http://www.ser.org/content/directory_of_restoration_expertise.asp

U.S. Department of Agriculture (USDA)
  APHIS Noxious Weeds Program:
  http://www.aphis.usda.gov/plant_health/plant_pest_info/weeds/index.shtml

  Forest Service Invasive Species Program:
  http://www.fs.fed.us/invasivespecies/index.shtml

USDA Plants Database:
http://plants.usda.gov/

U.S. Fish and Wildlife Service
http://www.fws.gov/invasives/

U.S. Geological Survey Biology: Invasive Species Program:
http://biology.usgs.gov/invasive/
USGS Nonindigenous Aquatic Species:
http://nas.er.usgs.gov/taxgroup/plants/

Wildlife Habitat Council – Invasive Species Program:
http://www.wildlifehc.org/managementtools/DODLegacy.cfm

Native Plant Resources

Audubon Center for Native Plants at Beechwood Farms Nature Reserve:
http://www.aswp.org/acnp.html

Bowman's Hill Wildflower Preserve
http://www.bhwp.org/

Delaware Native Plant Society:
http://www.delawarenativeplants.org/dnps-pagetwo.htm

Lady Bird Johnson Wildflower Center:
http://www.wildflower.org/

Mid-Atlantic Native Plant Sales, U.S. EPA:
http://www.epa.gov/reg3esd1/garden/lsales.htm

Native Plant Society of New Jersey:
http://www.npsnj.org/

Pennsylvania Native Plant Society:
http://www.pawildflower.org/
Nursery List: http://www.pawildflower.org/04_links/links2.htm

The Pennsylvania Flora Project - Morris Arboretum, University of Pennsylvania:
http://www.paflora.org/

Plant Conservation Alliance:
http://www.nps.gov/plants/index.htm

U.S. Department of Transportation: Roadside Use of Native Plants:
http://www.fhwa.dot.gov/environment/rdsduse/index.htm

REGIONAL PARTNERSHIPS

Delaware River Basin Commission
http://www.state.nj.us/drbc/

DOD RESOURCES

Armed Forces Pest Management Board:
http://www.afpmb.org/

Center for Environmental Management of Military Lands:
http://www.cemml.colostate.edu/

Defense Environmental Network and Information eXchange (DENIX):
https://www.denix.osd.mil/

Defense Technical Information Center (DTIC):
http://www.dtic.mil/

Defense Reutilization and Marketing Service:
http://www.drms.dla.mil/

Joint Military Services Chesapeake Bay Program:
http://www.hqda.army.mil/acsim/env/cbi/

U.S. Army Corps of Engineers (USACE) Engineer and Research Development Center (ERDC):
http://www.erdc.usace.army.mil/

Noxious and Nuisance Plant Management Information System (PMIS):
http://el.erdc.usace.army.mil/pmis/

Aquatic Plant Information System (APIS):
http://el.erdc.usace.army.mil/aqua/apis/

U.S. Army Environmental Command (USAEC):
http://aec.army.mil/usaec/

PESTICIDE INFORMATION

The Extension Toxicology Network (EXTOXNET):
http://extoxnet.orst.edu/

Kellysolutions Service:
http://www.kellysolutions.com/

National Pesticide Information Center:

http://npic.orst.edu/

North American Pollinator Protection Campaign – Reducing Risk to Pollinators:
http://www.nappc.org/PesticidesMain.html

Pesticide Action Network (PAN) Pesticide Database:
http://www.pesticideinfo.org/Index.html

U.S. Environmental Protection Agency (EPA) – Pesticides:
http://www.epa.gov/pesticides/index.htm
ECOTOX Database:
http://cfpub.epa.gov/ecotox/

Virginia Tech Pesticide Programs:
http://www.vtpp.ext.vt.edu/

Information Sources

American Sheep Industry Association (ASI). 2006. Targeted Grazing: A Natural Approach to Vegetation Management and Landscape Enhancement. http://www.cnr.uidaho.edu/rx-grazing/Handbook.htm. (Accessed May 23, 2007).

Averill, K.M., DiTommaso, A., & S.H. Morris. 2008. Response of pale swallow-wort (Vincetoxicum rossicum) to triclopyr application and clipping. Invasive Plant Science and Management 1: 196-206.

Babbitt, B. 1998. Statement by Secretary of the Interior on invasive alien species. Proceedings, National Weed Symposium, BLM Weed Page. April 8-10. *In:* Pimentel, D., R. Zuniga, and D. Morrison. 2004. Update on the environmental and economic costs associated with alien-invasive species in the United States. Ecological Economics.

Berkowitz, A.R., M. Archie, and D. Simmons. 1997, Defining environmental literacy: a call to action. Bulletin of the Ecological Society of America.78:170-172.

Brooks, M.L., C.M. D'Antonio, D.M. Richardson, J.B. Grace, J.E. Keeley, J.M. DiTomaso, R.J. Hobbs, M. Pellant, and D. Pyke. 2004. Effects of invasive alien plants on fire regimes. BioScience 54:677-688. *In:* DiTomaso, J.M., and D.W. Johnson (eds.). 2006. The Use of Fire as a Tool for Controlling Invasive Plants. Cal-IPC Publication 2006-01. California Invasive Plant Council: Berkeley, CA. 56 pp.

Casagrande, R.A. & J.E. Dacey. 2007. Monarch butterfly oviposition on swallow-worts (Vincetoxicum spp.). Environmental Entomology 36: 631-636.

Center for Environmental Management of Military Lands (CEMML). *Wildland Fire.* http://www.cemml.colostate.edu. Colorado State University.

Clark, K.H. 1998. Use prescribed fire to supplement control of an invasive plant, Phragmites australis, in marshes of southeast Virginia. *In:* DiTomaso, J.M., and D.W. Johnson (eds.). 2006. The Use of Fire as a Tool for Controlling Invasive Plants. Cal-IPC Publication 2006-01. California Invasive Plant Council: Berkeley, CA. 56 pp.

DAIM-ED-N (200-3). June 26, 2001. Army Policy Guidance: Management and Control of Invasive Species. United States Department of Defense, Department of the Army, Assistant Chief of Staff for Installation Management. Washington, D.C.

Darcy, A.J., & M.C. Burkart. 2002. Allelopathic potential of Vinca minor, an invasive exotic plant in west Michigan forests. Bios 73(4): 127-132.

Denoth, M., L. Frid, and J. H. Myers. 2002. Multiple agents in biological control: improving the odds? Biological Control 24: 20-30.

Delaware River Basin Commission. 2008. State of the basin report. http://www.state.nj.us/drbc/SOTB/index.htm.

Department of Defense Wildland Fire Policy Working Group. 1997. Final Report. https://www.denix.osd.mil/denix/Public/Library/OSH-Fire/fire.html.

DiTommaso, A. & J.E. Losey. 2003. Oviposition preference and larval performance of monarch butterflies (Danaus plexippus) on two invasive swallow-wort species. Entomologia Experimentalis et Applicata 108: 205-209.

DiTommaso, J.M., and D.W. Johnson (eds.). 2006. The Use of Fire as a Tool for Controlling Invasive Plants. Cal-IPC Publication 2006-01. California Invasive Plant Council: Berkeley, CA. 56 pp.

DiTommaso, J.M., M.L. Brooks, E.B. Allen, R. Minnich, P.M. Rice, and G.B. Kyser. 2006. Control of invasive species with prescribed burning. Weed Technology 20:535-548.

DOD 4500.9-R. Defense Transportation Regulation – Part V. Department of Defense Customs and Border Clearance Policies and Procedures. United States Transportation Command (http://www.transcom.mil/j5/pt/dtr.cfm). September 29, 2006.

DOD Partners in Flight. 2009. Checklist of birds Naval Support Facility Indian Head, Maryland. http://www.dodpif.org/checklists/indianhead.htm.

Ehler, L.E., and R.W. Hall. 1982. Evidence for competitive exclusion of introduced natural enemies in biological control. Environmental Entomology. 40: 297-331. *In:* Denoth, M., L. Frid, and J. H. Myers. 2002. Multiple agents in biological control: improving the odds? Biological Control 24: 20-30.

Executive Order (EO) 13112 of February 3, 1999. Invasive Species. Washington, D.C. The White House. Federal Register Vol. 64 No. 25, February 8, 1999. http://www.invasivespeciesinfo.gov/laws/execorder.shtml

Gelbard, J.L. and J. Belnap. 2003. Roads as conduits for exotic plant invasions in a semiarid landscape. Conservation Biology 17(2):420-432.

Glass, W.D. 1991. Vegetation management guideline: cut-leaved teasel (*Dipsacus laciniatus* L.) and common teasel (*Dipsacus sylvestris* L.). Natural Areas Journal. 11:213-214. *In:* Ditomaso, J.M., M.L. Brooks, E.B. Allen, R. Minnich, P.M. Rice, and G.B. Kyser. 2006. Control of invasive species with prescribed burning. Weed Technology. 20:535-548.

Goodman, S.W. 1996. Ecosystem Management at the Department of the Defense. Ecological Applications 6(3): 706-707.

Gover, A., J.M. Johnson, and L.J. Kuhns. 2003. Roadside Vegetation Management Report – Sixteenth Year Report. The Commonwealth of Pennsylvania Department of Transportation. The Pennsylvania State University. http://vm.cas.psu.edu/2002/2002PDF/AR02_Final.pdf (Accessed July 26, 2007)

Greenberg, C.H., S.H. Crownover, and D.R. Gordon. 1997. Roadside soil: a corridor for invasion of xeric scrub by nonindigenous plants. Natural Areas Journal 17:99-109. *In*: Gelbard, J.L. and J. Belnap. 2003. Roads as conduits for exotic plant invasions in a semiarid landscape. Conservation Biology 17(2):420-432.

Hoyer, M.V. and D.E. Canfield, Jr. (eds.). 1997. Aquatic Plant Management in Lakes and Reservoirs. Prepared by the North American Lake Management Society (Madison, WI) and the Aquatic Plant Management Society (Lehigh, FL) for U.S. Environmental Protection Agency, Washington, D.C. http://plants.ifas.ufl.edu/hoyerapm.html (Accessed July 18, 2007).

Kenny, D.S. 1999. Historical and sociopolitical context of the western watersheds movement. Journal of the American Water Resources Association 35(3): 493-503.

Leach, W.D., N.W. Pelkey, and P.A. Sabatier. 2001. Stakeholder Partnerships as an Emergent Form of Collaborative Policymaking: Evaluation Criteria Applied to Watershed Management in California and Washington. Journal of Policy Analysis and Management.

Ludke, L., F. D'Erchia, J. Coffelt, and L. Hanson. 2002. Workshop Report – Office of the Regional Biologist, Central Region: Invasive Plant Species Inventory, Mapping, and Monitoring – A National Strategy. U.S. Geological Survey BRD Information and Technology Report ITR 2002-0006. U.S. Department of the Interior, U.S. Geological Survey.

Mack, R.N. and W.M. Lonsdale. 2001. Humans as global plant dispersers: getting more than we bargained for. BioScience 51(2): 95-102.

Matilla, H.R. & G.W. Otis. 2003. A comparison of the host preference of monarch butterflies (*Danaus plexippus*) for milkweed (Asclepias syriaca) over dog-strangler vine (*Vincetoxicum rossicum*). Entomologia Experimentalis et Applicata 107: 193-199.

McEvoy, P.B. and E.M. Coombs. 2000. Why things bite back: unintended consequences of biological weed control. Pp. 167-194 *In:* P.A. Follett and J.J. Duan (eds.) Nontarget effects of biological control. Kluwer Academic Publishers. Boston, Massachusetts.

Mehrhoff, L.J., J.A. Silander, Jr., S.A. Leicht, E.S. Mosher and N.M. Tabak. 2003. IPANE: Invasive Plant Atlas of New England. Department of Ecology & Evolutionary Biology, University of Connecticut, Storrs, CT, USA. URL: http://www.ipane.org

Memorandum of Understanding (MOU) Between the U.S. Environmental Protection Agency and the U.S. Department of Defense with Respect to Integrated Pest Management. 1996. (Accessed June 5, 2007) https://www.denix.osd.mil/denix/Public/ES-Programs/ES/Documents/note2.html

Michaels, S., R.J. Mason, and W.D. Solecki. 1999. The importance of place in partnerships for regional environmental management. Environmental Conservation 26(3): 159-162.

Miller, J.H. 2003 Nonnative Invasive Plants of Southern Forests: a field guide for identification and control. Revised. Gen. Tech. Rep. SRS-62. Asheville, NC: U.S. Department of Agriculture, Forest Service, Southern Research Station. 93 p.

Mueller, T.C., D.K. Robinson, J.E. Beeler, C.L. Main, D. Soehn, and K. Johnson. 2003. *Dioscorea oppositifolia* L. phenotypic evaluations and comparison of control strategies. Weed Technology 17:705-710.

Murphy, K.J. 1988. Aquatic weed problems and their management: a review. II. Physical control measures. Crop Protection 7:283-302. AFPMB LRS (Accessed July 17, 2007)

Murphy, S.D., J. Flanagan, K. Noll, D. Wilson, and B. Duncan. 2007. How incomplete exotic species management can make matters worse: experiments in forest restoration in Ontario, Canada. Ecological Restoration 25(2)85-93.

National Invasive Species Council (NISC). 2001. Meeting the Invasive Species Challenge: National Invasive Species Management Plan. 80pp.

National Research Council. 2002. Predicting invasions of nonindigenous plants and plant pests. Washington, D.C. National Academy Press. *In:* Reed, C.C. 2004. Keeping invasive plants out of restorations. Ecological Restoration 22(3): 210-216.

NatureServe. 2006. NatureServe Explorer: An online encyclopedia of life [web application]. Version 6.1. NatureServe, Arlington, Virginia. Available http://www.natureserve.org/explorer. (Accessed: March 15, 2007)

Nealen, H.J. & P. M. Nealen. 2000. Ruby-Throated Hummingbird Death by Common Burdock (Arctium minus). The Wilson Bulletin 112(3): 421-422.

Neary, D.G., C.C. Klopatek, L.F. DeBano and P.F. Ffolliott. 1999. Fire effects on belowground sustainability: a review and synthesis. Forest Ecology and Management 122(1-2):51-71. *In*: DiTomaso, J.M., and D.W. Johnson (eds.). 2006. The Use of Fire as a Tool for Controlling Invasive Plants. Cal-IPC Publication 2006-01. California Invasive Plant Council: Berkeley, CA. 56 pp.

Olson, L.J., and S. Roy. 2002. The economics of controlling a stochastic biological invasion. American Journal of Agricultural Economics. 84 (5):1311-1316

Paciulli, Simmons and Associates. 2006. Invasive Exotic Vegetation Management Plan Fort Belvoir, Virginia. Fairfax, VA.

Parker, P.A. Memorandum: FY 96 Report - DOD Pest Management Measures of Merit. Department of the Air Force, Headquarters Air Combat Command, Langley Air Force Base, Virginia. (Accessed May 23, 2007). https://www.denix.osd.mil/denix/Public/Policy/AF/Policy/note17.html.

Pimentel, D., R. Zuniga, and D. Morrison. 2004. Update on the environmental and economic costs associated with alien-invasive species in the United States. Ecological Economics.

Plant Conservation Alliance, Alien Plant Working Group. Least Wanted: Alien Plant Invaders of Natural Areas Fact Sheets. http://www.nps.gov/plants/alien/factmain.htm

Plant Conservation Alliance. Federal Native Plant Conservation Memorandum of Understanding (MOU). http://www.nps.gov/plants/mou.htm (Accessed August 12, 2007)

Reed, C.C. 2004. Keeping invasive plants out of restorations. Ecological Restoration 22(3): 210-216.

Rice, P.M. 2005. Fire as a tool for controlling nonnative invasive plants. www.weedcenter.org/management/tools.htm#burning. Bozeman, MT: Center for Invasive Plant Management.

Richardson, D.M. 2004. Plant invasion ecology –dispatches from the front line. Diversity and Distributions, 10:315-319.

Richburg, J.A. and W.A. Patterson III. 2003. Can northeastern woody invasive plants be controlled with cutting and burning treatments? *In:* Ditomaso, J.M., M.L. Brooks, E.B. Allen, R. Minnich, P.M. Rice, and G.B. Kyser. 2006. Control of invasive species with prescribed burning. Weed Technology. 20:535-548.

Schmidt, W. 1989. Plant dispersal by motor cars. Vegetation 80:147-152. *In:* Gelbard, J.L. and J. Belnap. 2003. Roads as conduits for exotic plant invasions in a semiarid landscape. Conservation Biology 17(2):420-432.

Sheley, R.L. and J. Krueger-Mangold. 2003. Principles for restoring plant-infested rangeland. Weed Science 51:260-265.

Simberloff, D. 2003. Eradication – preventing invasions at the outset. Weed Science, 51:247-253.

Simberloff, D. and P. Stiling. 1996. Risks of species introduced for biological control. Biological Conservation, 78:185-192.

Southeast Exotic Pest Plant Council (SE-EPPC). Invasive Plant Manual. http://www.se-eppc.org/manual/index.hmtl

Tilman, D. 1986. Resources, competition and the dynamics of plant communities. Pages 51-75 in Michael Crawley, ed. Plant Ecology. Boston: Blackwell Scientific. *In:* Sheley, R.L. and J. Krueger-Mangold. 2003. Principles for restoring plant-infested rangeland. Weed Science 51:260-265.

Trauger, D.L., W.C. Tilt, and C.B. Hatcher. 1995. Partnerships: Innovative strategies for wildlife conservation. Wildlife Society Bulletin 23(1): 114-119.

Toupal, R.S. and M.D. Johnson. Conservation Partnerships: Indicators of Success. Social Sciences Institute, Technical Report. Release 7.1 (February 1998).

Trombulak, S.C. and C.A. Frissell. 2000. Review of ecological effects of roads on terrestrial and aquatic communities. Conservation Biology 14:18-30. *In*: Gelbard, J.L. and J. Belnap. 2003. Roads as conduits for exotic plant invasions in a semiarid landscape. Conservation Biology 17(2):420-432.

Tu, M., C. Hurd, and J.M. Randall. 2001. Weed Control Methods Handbook, The Nature Conservancy, http://tncweeds.ucdavis.edu, version: April 2001.

United States Army Corps of Engineers, Engineer Research and Development Center. Noxious and Nuisance Plant Management Information System (PMIS). Version 5.3. Vicksburg, MS.

United States Department of Agriculture, Forest Service. Weeds of the Week. http://www.na.fs.fed.us/fhp/invasive_plants/weeds/index.shtm

United States Environmental Protection Agency, Office of Policy, Planning, and Evaluation. 1992. Enhancing Management of Forests and Vegetation on Department of Defense Lands: Opportunites, Benefits, and Feasibility. Prepared by The American Forestry Association – Clark Row, Principal Investigator.

Van Driesche, R., S. Lyon, B. Blossey, M. Hoddle, and R. Reardon. 2002. United States Department of Agriculture Forest Service. Biological Control of Invasive Plants in the Eastern United States. Publication FHTET-2002-04. Morgantown, West Virginia.

Virginia Invasive Species Council (VISC). 2005. Virginia Invasive Species Management Plan. http://www.dcr.virginia.gov/natural_heritage/vaisc/documents/VISMP_final.pdf

Vitousek, P.M., C.M. D'Antonio, L.L. Loop, and R. Westbrooks. 1996. Biological invasions as global environmental change. American Scientist 84: 468-478. *In:* Mack, R.N. and W.M. Lonsdale. 2001. Humans as global plant dispersers: getting more than we bargained for. BioScience 51(2): 95-102.

Weeks, Sally S. 2003. Red and White Mulberry in Indiana. Purdue University Forestry and Natural Resources Know your Trees Series. http://www.ces.purdue.edu/extmedia/FNR/FNR_237.pdf.

Westbrook, C., K. Ramos and M. La. 2005. Under Siege: Invasive Species on Military Bases. National Wildlife Federation. http://www.nwf.org/.

Wilcove, D.S., Rothstein, D., Dubow, J., Phillips, A. & E. Losos. 1998. Quantifying threats to imperiled species in the United States: Assessing the relative importance of habitat destruction, alien species, pollution, overexploitation and disease. BioScience 48(8): 607-615.

Glossary of Plants

CLEARANCE REQUEST FOR PUBLIC RELEASE OF DEPARTMENT OF DEFENSE INFORMATION

(See Instructions on back.)

(This form is to be used in requesting review and clearance of DoD information proposed for public release in accordance with DoDD 5230.9.)

TO: *(See Note)* **Chief, Office of Security Review, 1155 Defense Pentagon, Washington, DC 20301-1155**

Note: Regular mail address shown above. For drop-off/next day delivery, use:
Room 12047, 1777 North Kent Street, Rosslyn, VA 22209-2133

1. DOCUMENT DESCRIPTION

a. TYPE Report	b. TITLE INVASIVE SPECIES GUIDEBOOK FOR DEPARTMENT OF DEFENSE INSTALLATIONS IN THE DELAWARE RIVER BASIN
c. PAGE COUNT 204	d. SUBJECT AREA Conservation

2. AUTHOR/SPEAKER

a. NAME *(Last, First, Middle Initial)* Boice, L. Peter	b. RANK NH IV	c. TITLE Conservation Team Leader
d. OFFICE ODUSD(IE)		e. AGENCY OSD

3. PRESENTATION/PUBLICATION DATA *(Date, Place, Event)*

Posting on the public side of DENIX https://www.denix.osd.mil & https://www.DoDLegacy.org, Please fax copy of cleared form to 703-607-3124 or email Pedro.Morales.ctr@osd.mil

4. POINT OF CONTACT

a. NAME *(Last, First, Middle Initial)* Boice, L. Peter	b. TELEPHONE NO. *(Include Area Code)* 703-604-0524

5. PRIOR COORDINATION

a. NAME *(Last, First, Middle Initial)*	b. OFFICE/AGENCY	c. TELEPHONE NO. *(Include Area Code)*
Morales-Muniz, Pedro J.	ODUSD(IE)-CO	703-604-1933

6. REMARKS

(Legacy 08-328) This reports provides a guide for invasive plant species identification, management techniques, how to guide on preventing recurring invasive species and restoring historical plant communities, forming cooperative partnerships to acheive management goals and case studies.

CLEARED
For Open Publication

OCT 13 2009 5

Office of Security Review
Department of Defense

7. RECOMMENDATION OF SUBMITTING OFFICE/AGENCY

a. THE ATTACHED MATERIAL HAS DEPARTMENT/OFFICE/AGENCY APPROVAL FOR PUBLIC RELEASE *(qualifications, if any, are indicated in Remarks section)* AND CLEARANCE FOR OPEN PUBLICATION IS RECOMMENDED UNDER PROVISIONS OF DODD 5320.9. I AM AUTHORIZED TO MAKE THIS RECOMMENDATION FOR RELEASE ON BEHALF OF:

b. CLEARANCE IS REQUESTED BY 20091015 *(YYYYMMDD)*.

c. NAME *(Last, First, Middle Initial)* Boice, L. Peter	d. TITLE Conservation Team Leader
e. OFFICE ODUSD(IE)	f. AGENCY OSD
g. SIGNATURE	h. DATE SIGNED *(YYYYMMDD)* 20090923

10-S-0046