Introduction

The National Park Service is proud to be a leader among federal agencies in the implementation of Integrated Pest Management (IPM). With over 80 million acres of land, 45,000 buildings and cultural landscapes ranging from croplands to historic rose gardens, we face every conceivable pest problem. Since implementing an IPM program in the early 1980's, the Park Service has reduced pesticide use by over 60 percent while improving the effectiveness of our pest management efforts. Key elements in this success were formal training and the provision of printed and audiovisual materials.

One of our products is an IPM Manual which is now available in a second edition. It provides descriptions of the biology and management of 21 species or categories of pests. The Park Service is pleased to offer this information to the IPM community.

The National Park Service wishes to thank the Entomology Department at Colorado State University. They designed the original NPS IPM Manual website and made it available on the Internet before the Park Service's natural resource website was fully operational.

Terry Cacek
IPM Coordinator

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Ants

This module is intended to serve as a source of basic information needed to implement an Integrated Pest Management program for structure-infesting ants. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

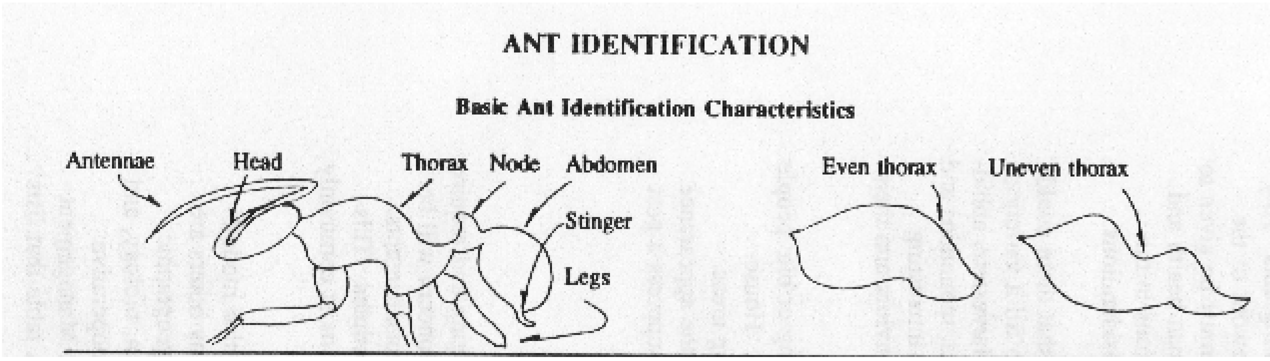
Ants are among the most successful insects. Experts estimate that there could be 20,000 or more species of ants in the world. They have evolved to fill a variety of different ecological niches as predators, herbivores, leaf-cutters, seed-harvesters, aphid- tenders, and fungus-growers. They are found in deserts and rainforests, mountains and valleys, from the Arctic Circle to the tip of South America. They are interesting organisms that should be studied to better understand their unique behaviors and their roles in the earth's ecosystems.

They can also be pests, however. Fire ants and others may sting or bite people and animals. Pharaoh ants get into wounds and dressings in hospitals. House-infesting ants can become pests by their presence in kitchens and living areas. Carpenter ants tunnel into structural wood. Mound-building ants mar the appearance of lawns and landscaped areas. Sometimes ants must be managed to suppress a pest problem.

IDENTIFICATION AND BIOLOGY OF ANTS

Only a comparatively small proportion of ant species are damaging and require control. For National Park Service personnel, the ants most often of concern will be species in three groups: fire ants, carpenter ants, and house-infesting nuisance ants. The first two are discussed in separate Integrated Pest Management modules. This module addresses the third group, house-infesting ants--those ants that most commonly invade structures looking for food, water, or nesting sites.

A detailed description of every pest ant is beyond the scope of this module. Well over a dozen are considered common pests of structures, and many others are occasional pests. The species most likely encountered will depend on geographic location and surrounding habitat. Detailed information on identification, biology, and management of specific pest ants should be obtained from the local Cooperative Extension Service, your regional National Park Service Integrated Pest Management coordinator, or from the References listed at the end of this module. A table that lists key features used to identify major pest species of ants follows.



Species	Worker Size	Color	Thorax Shape	No. of Nodes	Other ID Characteristics	Treatment	Bait
Pavement ant	3/16”	Dark brown	Uneven	2	Grooves on head +thorax stinger at tip of abdomen	Barrier-poor nest-excellent	Sweet
Thief ant	1/32”	Yellow	Uneven	2	Stinger at tip of abdomen	Barrier-poor	Sweet or protein
Crazy ant	1/8”	Dark brown	Uneven	One	Very long legs. First antennal segment twice as long as head.	Barrier-good Nest-excellent	Sweet or protein
Field ant	3/8”	Brown to very dark brown	Uneven	One	None	Barrier-excellent	Sweet
Pharaoh ant	1/16”	Yellow with red abdomen	Uneven	One	None	Barrier-poor	Sweet or protein
Argentine Ant	1/8”	Brown; sometimes light brown	Uneven	One	Sparse body hairs; no hairs on thorax	Barrier-poor	sweet

The Ant Colony and Life Cycle

Ants belong to the insect order Hymenoptera, which also includes the wasps and bees. Ants are distinguished from many of their nearest relatives by two characteristics: a narrow "waist" (the slender free-moving portion of the abdomen called a **pedicel**) and elbowed antennae.

Ants also differ from most other insects in that they are social, similar to termites and certain bees and wasps. This means that ants live in large cooperative groups called colonies. Two or more generations overlap in the colony; adults take care of the young and are divided into castes, specialized groups that take care of certain tasks. Ants have reproductive castes, the queens and males, and nonreproductive castes, the workers.

Queens. A queen is generally the largest individual in the colony. She has wings until after her mating flight, when she removes them. The primary function of the queen is reproduction, but after establishing a new nest she may also care for and feed the first brood of workers. Once she has produced her first brood, she becomes an "egg-laying machine," cleaned and fed by her offspring. She may live for many years until replaced by a daughter queen. Some ant species have more than one queen in the nest.

Males. Male ants are generally winged and usually keep their wings until death. Apparently, the male ant's only function is to mate with the queen. Once he does, he dies, generally within two weeks. Males are produced in old, mature colonies.

Workers. The workers are sterile, wingless females who build and repair the nest, care for the brood, defend the nest, and feed both immature and adult ants, including the queen. There may be workers and soldiers of different sizes that specialize in certain tasks.

Ants develop through a complete life cycle of egg, larva, pupa, and adult. The egg is tiny, almost microscopic in size. The larva is legless and grub-like, very soft and whitish in color. It is also helpless and depends totally on workers for food and care. The pupa looks somewhat like the adult but is soft, unpigmented, and cannot move around. Some are enclosed in a cocoon, some are not. A newly-emerged adult requires several days for its body to harden and darken.

New Colony Formation

Once a colony of ants matures, it can establish new colonies through various methods, depending on the species. The two most common are budding and swarming. The appropriate management strategy depends on how a colony spreads, so it is essential to correctly identify the ant species before deciding how to manage it.

Budding. Budding is the breakaway of a group of ants from a mature colony to form a new colony. The group usually consists of one or more queens and some workers carrying larvae. Budding is common with species of ants that have multiple queens, such as Pharaoh ants and Argentine ants. Residual insecticides should **not** be used for ants that undergo budding because they can stimulate this process.

Swarming. Most ants establish new colonies through swarming. Every now and then, particularly in spring or early summer, mature ant colonies generate large numbers of winged forms. These are the young queens and males, going off to mate. An inseminated queen then rids herself of her wings and attempts to start a new nest in a cavity, under a stone or a piece of bark, or by excavating a hole in the ground. She rears her first brood alone, feeding them with salivary secretions and infertile eggs. If successful, the first brood opens up the nest and brings in food for themselves, the queen, and subsequent broods, and the colony grows. However, the percentage of queens that successfully begin new colonies is thought to be very small.

The Difference Between Winged Ants and Winged Termites

Although ants and termites are very different, they are often confused. They are alike in that they live in colonies and periodically swarm. Swarming forms of both are dark and winged. But worker termites are whitish and never seen running freely about as do ants. Instead, termites remain protected in their nests and galleries in wood and soil.

Winged adult ants can be told from winged termites by the following differences. Winged ants have a narrow waist, front wings that are larger than the rear, and elbowed antennae. Winged termites have a fat waist, equally sized wings, and straight, beaded antennae.

Seasonal Abundance

Most outdoor ants increase in population and activity from spring into summer months and then decline from fall into early winter as the temperature drops and the ants' natural food supplies dwindle. Other ants, such as the Argentine ant, may increase in numbers in the fall as various colonies aggregate together to overwinter. Some ants, such as the Pharaoh ant, which may live entirely indoors, exhibit little seasonality.

Feeding Habits

Knowing the food habits of the particular ant species is important in ant management because it may enable the location and elimination of the food that is attracting the ants to the site, it can help to locate foraging trails to track the ants back to their nest, and it can help to choose an effective bait.

Ants feed on many different types of food. Some species will feed on practically anything; others may limit their food to a narrow range. Ants infesting structures are typically feeding on "people food," both food in storage (sugar, cakes, cookies, breakfast cereals, etc.) and food from spills and garbage. But they may also be preying on other insects or scavenging on dead insects in windows or lights.

Food preferences are often seasonal. When the queen is actively laying eggs, worker ants typically gather protein- based foods for the queen. At other times they may ignore protein foods completely and restrict their foraging to sugars and greases.

Many ants obtain sugar by feeding on honeydew, a sweet substance secreted by aphids and other plant-sucking insects. They often defend these insects from predators and tend them as if they were their personal food supply. Indoor infestations of ants are occasionally traced to large populations of aphids on outdoor foundation plants or indoor houseplants.

The six most common ant species that infest buildings are the pavement ant, the thief ant, the crazy ant, the field ant, the Pharaoh ant, and the Argentine ant.

Pavement Ant Identification and Biology

Pavement ants (*Tetramorium caespitum*) were introduced to the United States from Europe and occur throughout the eastern United States. They are an important pest in the midwest and New England. These are small ants, about 3/16" long, and are dark brown in color. They build nests along sidewalks, building foundations, and under stones, boards, bricks, and mulch or leaf piles. These ants readily make trails to and from food sources and often forage along the edge of carpeting or baseboards. They are also common around the base of toilets. They often nest in protected areas so the nests may be hard to locate, but this is essential to manage infestations of this species. There can be several thousand in a colony.

Pavement ants feed on a wide variety of foods including other insects, greasy foods, and plants. While they are often found in damp areas, lack of moisture does not limit their development, so solving moisture problems alone will not affect these ants. Vegetation-free borders should be installed around buildings, and any cracks in building foundations should be sealed. Any loose material under that could provide nesting habitats and should be raised off the ground.

Thief Ant Identification and Biology

The native thief ant (*Solenopsis molesta*) is found throughout the United States, but primarily in the eastern and central states. A very small ant, thief ants are easily confused with the Pharaoh ant. The best way to tell them apart is to look at the club on the end of the antenna with a magnifying glass and count the number of segments; thief ants have two segments, while Pharaoh ants have three. Thief ants are named for their habit of stealing food from the nests of other ants. They nest outside under debris, rocks, or logs; indoors they nest in wall voids and behind baseboards. They are very small and can easily enter packaged foods, so food should be enclosed in tightly-sealed containers. Locating thief ants' nests can be difficult and time-consuming because their small size can make it difficult to follow the trail. Thief ants feed on both protein and sweets and will tend aphids, mealybugs, and scales to obtain the honeydew they excrete.

All cracks in walls should be sealed to keep these ants from entering buildings. Patience is essential in managing the ants because the nest can be so hard to locate. Baits do not seem to be effective for thief ants since they tend not to eat enough bait to bring sufficient quantities back to the nest for it to work.

Crazy Ant Identification and Biology

Crazy ants (*Paratrechina longicornis*) were introduced to the United States from India. Their distribution is limited to the Gulf coast from Florida to Texas. They are easily identified by their long legs and their habit of erratically moving from place to place (hence the name "crazy"). Crazy ant trails are not readily obvious because of this erratic movement. The easiest way to find the nest is to look for workers carrying pieces of food or workers with swollen abdomens. These ants are carrying food back to the nest. By observing their movement, it should be possible to find the nest. Crazy ants are highly adaptable and will nest in a variety of locations, from very dry to moist. They will nest under objects, in rotten wood or trash, in tree cavities, as well as in debris left standing in buildings for long periods of time.

These ants feed on a variety of foods including grease, sweets, and other insects. In some areas they are considered a biological control agent for houseflies. They also tend aphids and scales to feed on their honeydew. While crazy ants need moisture, elimination of water by itself will not get rid of these ants since they can survive under a wide range of conditions. Elimination of food sources and nest sites are equally important in the management of this ant.

Crazy ants do not respond well to baits, so they cannot be relied upon for management of this ant. Surrounding buildings with vegetation-free barriers such as stone or brick (but not wood mulch) will keep ants from entering buildings to nest.

Field Ant Identification and Biology

Field ants (*Formica* spp.) are found throughout the United States but primarily in the Midwest and North. They are large (3/8" long) and dark brown to black. They are often confused with the carpenter ant, but can be distinguished by an uneven thorax (see ant identification chart at the end of this module). Field ants feed on other insects as well as insect honeydew. They cause concern because they usually nest near structures and are often mistaken for carpenter ants. Nests are often made in grassy areas and can be difficult to see because they are low to the ground. Field ants will also nest in leaf litter or mulch that is more than two inches thick, and can live under stones, firewood, or other debris that might be found in a lawn area. If pesticide drenches of mounds are used to manage this insect it should be remembered that they will be slow to act because it often takes foraging ants days to return to the nest.

Pharaoh Ant Identification and Biology

Pharaoh ants, (*Monomorium pharaonis*), are small yellow ants about 1/16" long. They are easily confused with thief ants, also a small yellow ant. To distinguish the two, it is necessary to look at the antennae. Pharaoh ants have twelve segments with a three-segmented club on the end, while thief ants have ten segments with a two-segmented club. Pharoah ants are native to tropical Africa but are now distributed throughout the world. They are usually associated with heated buildings since they cannot survive outside year round in the majority of the United States. These ants will nest in any dark void in a structure as well as in folded bags or newspapers. In the subtropical United States they will nest outside in leaf litter, piles of bricks, potted plants, or under roof shingles.

Pharaoh ant colonies can become quite large, often containing as many as 300,000 workers with several queens. New colonies are formed by budding, when some of the workers, brood, and a few queens move to a new location. In warm areas where they can survive outdoors they will move from building to building.

Pharaoh ant management is more dependant on locating areas of ant activity than eliminating the colony, since they are so large and can spread so easily. Place jelly baits on 1" squares of paper or tape and place in damp, dark areas. These ants move along electrical wires, so an inspection should include areas where wires enter walls or appliances, as well as behind switchplates and outlets. Pharoah ants will also nest in and around appliances such as refrigerators or stoves that have food or water around them. A useful tool for the management of this ant is to make a map of the site and mark locations where ants and their colonies are found. This will help to identify new areas of activity over time.

Sanitation is essential for Pharaoh ant management, since elimination of food sources will make them more receptive to insecticide baits. Residual insecticides should not be used for Pharaoh ant management. They can repel ants, forcing more colonies to form through budding while killing only a small number of ants. During the first two to four weeks of the program, place baits containing an insect growth regulator and a food attractant inside a soda straw throughout the area of infestation. These should be located along edges and in corners where ants are most likely to encounter them. Placing baits inside straws will keep them fresh and away from people and domestic animals. Replace these with boric acid/food attractant baits. One food bait is three parts honey: two parts peanut butter: one part mint apple jelly : one part egg yolk baby food. Commercial baits are also available. Exterior treatments may be necessary in subtropical areas of the United States or during the warmer months in northern areas. Remember that both insect growth regulators and boric acid are EPA- registered pesticides, so your regional National Park Service Integrated Pest Management coordinator should be consulted before using these materials.

Argentine Ant Identification and Biology

Argentine ants, (*Iridomyrmex humilis*), are an imported species common throughout the southeast and southern California. These ants will nest in soil and mulch, as well as under stones, logs, and debris. They are often found in tree holes, bird nests, leaf litter, and bee hives. These ants form large colonies; workers from different colonies do not fight and will often join together to form larger colonies. This means that areas from which colonies are eliminated can quickly be repopulated. These large colonies will often split by budding during the warmer months. Although Argentine ants form winged reproductives, they do not swarm. They feed on a variety of foods but seem to prefer sweets and will feed on aphid honeydew. They will even feed on fruit crops and are considered an agricultural pest in some areas.

Argentine ant trails are easy to locate along sidewalks, foundations, and along the edges of buildings. If grass grows to the edge of the building it should be pulled back during an inspection. These ants will also move into buildings by climbing up trees onto wires entering buildings, so any place where branches touch buildings should be inspected as well. As with so many other ants, use of a vegetation-free border and correction of moisture problems will help in management of Argentine ants. Insecticide baits are useful for managing this ant.

MONITORING AND THRESHOLDS

Identification of the species will help to determine where the nest might be located, what the ants might be feeding on, and the best tactics for control. All parts of the building and the surrounding area should be inspected for ant activity as well as food and water sites. People that work in the building might have seen the ants also. Some species are most active in the evening. For these, a daytime inspection might discover little, while significant ant activity might be observed at midnight.

Some infestations may require an intensive survey program using nontoxic baits to determine likely nesting sites. Good baits are jelly, honey, peanut butter, bacon grease, or raw liver. The baits (or a combination of baits) should be placed on small pieces of cardboard, aluminum foil, masking tape, or plastic pill bottle lids throughout the building and periodically checked for feeding ants. Active sites should be noted on a survey diagram. Baits that haven't had any feeding activity in 24 hours should be moved. Over a period of days the survey diagram will pinpoint areas of activity. In addition, trails of ants feeding on the bait can sometimes be followed back to the nest site.

There is no single threshold level for house-infesting ants. Threshold levels need to be set separately for each site. For example, a single ant in a first-aid station may be one too many. In an eating area, control actions might be initiated if there were more than a half-dozen ants in a day, while most people's tolerance for ants in a rustic and open recreation room would likely be much higher.

NON-CHEMICAL CONTROL OF ANTS

The most effective ant control results from the destruction of the queens and the nest itself. If the nest is found by tracking workers, or through a survey, eliminating that nest is fairly simple, particularly if it is located, as it often is, outdoors, or in the soil beneath a cracked floor. It is simply a matter of mechanically destroying the nest.

But effective ant management is rarely that simple. Sometimes you can't find the nest. Often there are multiple nests. (One species, the Pharaoh ant, can have hundreds of small nests within a single room.) There may be a constant pressure from ant colonies invading from surrounding areas. In most cases, long-term management of pest ants means integrating improved sanitation, structural repairs, and habitat modification along with one or more direct control tactics such as insecticide baits, crack and crevice treatments, and direct physical controls.
Successful ant management usually requires a combination of management tactics, ranging from caulking to cleanup, improved sanitation to habitat modification, as well as targeted and limited insecticide treatment.

The keys to success in ant management are, first, vigorous inspection to determine the nature and extent of the infestation, and, if at all possible, the location of the nest. Second, meticulous sanitation to eliminate readily available food and water. Third, the choice of the right combination of tools to eliminate the problem. The listing for each ant species provides more information on management strategies relevant to that ant.

Improved Sanitation

Like all pests, ants need food, water, and shelter to survive. By limiting these three essentials, you make it more difficult for ants to live in the infested area. Simply by improving sanitation you can often suppress existing populations and discourage new invasions.

Ants can enter many types of food packaging, particularly once the package has been opened. (They have even been found inside glass jars after traveling around the threads of a screw-on lid!) Cereals, sugar, and other bulk food should be stored in plastic containers with snap-on lids, in glass jars with rubber seals, or in a refrigerator.

Food spills also feed ants. As with cockroaches, enthusiastic cleaning helps to minimize ants. Frequent vacuuming, sweeping, or mopping of floors and washing of counter and table tops eliminates much of the food ants may be foraging on. Trash should be stored away from infested areas and monitored for spills.

Ants can get their water from many sources inside a structure: condensation on pipes and air conditioners, leaky plumbing, aquariums, pet dishes, houseplant containers, floor drains, etc., and limiting these is rarely practical.

Ant-Proofing

Ants can enter and move through a structure through innumerable tiny cracks and openings. Yet caulking and otherwise sealing cracks and crevices being used by ants can often have great effect in suppressing the population. Many easy-to-use and effective silicon sealers and expandable caulk products have been recently developed, including some designed specifically for pest management. Repairing torn screens and installing doorsweeps can also prevent ants from easily entering a structure. Non-vegetation barriers such as stones or brick walkways next to a building can be helpful in helping to keep ants out of structures as well.

Habitat Modification

Trim the branches of trees located close to structures so the branches do not act as runways from nest sites to roof or siding. Alter landscaping to minimize the number of aphids and other honeydew-producing insects that attract ants. Firewood kept indoors should be moved outdoors or regularly inspected for ants. Don't stack wood next to structures and move trash, since ants often nest under objects. Moisture accumulation in buildings can also result in ant infestations.

Direct Physical Control

Ants can be discouraged from foraging in certain limited sites with sticky barriers. For example, commercially available sticky repellents or petroleum jelly can be applied in a narrow band around table legs to prevent ants from walking up to the tabletop. Double-sided tape can also be used.

Large numbers of worker ants can be mopped or sponged up with soapy water. Water, especially boiling water, has also been used to flood ant nests. Some ground-ant nests have been destroyed by digging them up and destroying the nest structure.

CHEMICAL CONTROL OF ANTS

Many people, on discovering ants, simply spray insecticide wherever they have seen ants. This is a poor strategy, usually doomed to failure. Applying undirected, general insecticide sprays indoors is unsatisfactory because the sprays only "harvest" a small portion of the workers and have little effect on the colony, the ultimate source of the problem. A further problem is that some species are apparently triggered into "budding" new colonies when they contact insecticide near their nests and foraging sites.

The chemical tools available for ant control have changed in the past few years with the addition of insect growth regulators, new baits, and commercial bait stations, and new tools can be expected in the future. Even so, insecticides are only one of the tools available for control of ants, and not always the best or most important. Ant biology should be considered when deciding whether or not to use insecticides. For example, insecticides are often not effective against mound ants because it often takes foraging ants several days to return to the nests. Consult your regional National Park Service Integrated Pest Management coordinator for information on using pesticides as part of an ant management program.

Ant baits. The best baits for ants are those whose toxicant kills ants slowly. In this way, worker ants live long enough to take the baits back to the nest and feed it to the colony and queen. A number of baits are now available. Some are prepackaged in child-resistant bait stations. Some are gels or pastes designed to be placed in small pea-shaped amounts throughout an area. Some products (such as boric acid) are designed to be mixed with a food. Bait products typically will work against certain species of ants but not against others, so it is important to check the label to make sure the ant you wish to control is listed.

Insect growth regulators (IGRs). These are available in bait form for some ant species. Insect growth regulators inhibit normal development of insects. They are slow-acting because they stop the next generation from developing rather than killing the current generation. A recent study comparing the insect growth regulator fenoxycarb to a commercial bait found that the growth regulator was more effective than the bait in eliminating Pharaoh ants. This is most likely because the bait kills ants too quickly to be effectively distributed throughout the colony (Williams and Vail 1994). Crazy ants do not seem to respond well to bait, and baits may be slow-acting against field ants since they often stay away from the nest for several days.

Liquid and aerosol insecticides. Nearly all of the insecticides labeled for use against cockroaches are also labeled for use against ants. These insecticides are most effective when used to treat actual nest sites. Insecticides are less effective, but still may provide acceptable results when used to treat inside cracks and crevices used by ants in and around infested sites. They are least effective, as well as offering the highest potential of human exposure, when they are simply applied to sites where activity has been observed.

Drenches. For certain ground-nesting ants that dig deep nests outdoors, a soil drench or mound drench can be effective where other treatments are not. As its name implies, a soil drench consists of applying enough insecticide dilution directly to a mound or nest so that the entire nest is drenched.

Dusts. Dusts may also be used on occasion for ant control if they are used lightly or directed into nests. In large amounts, dusts tend to repel ants. But they have the advantage of floating back through wall voids to reach nests that may not be accessible with other formulations.

Granules. Granules are rarely used in household ant control. They may be useful, however, when a lawn or field is heavily infested with many colonies of a shallow, ground- nesting species of ant.

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Aphids

This module is intended to serve as a source of basic information needed to implement an Integrated Pest Management program for aphids. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

The term aphid is applied to a large number of species of small, soft-bodied insects of the superfamily Aphidoidea, order Homoptera. The majority of aphid problems likely to be encountered within the National Park Service are caused by species in three families: the Aphididae (true aphids or plantlice), the Adelgidae (pine and spruce adelgids), and the Phylloxeridae (phylloxerans). These families comprise thousands of species and include some of the most important plant pests in the world.

Aphids vary greatly in their patterns of reproduction, use of hosts, and types of damage that they cause. In a single season some species may produce sexual and asexual forms. In other species sexual forms are unknown. Some species remain on the same host throughout the entire year and others may have obligatory alternation between two different hosts. Because of the large number of species and the variation which occurs within species, it is recommended that aphid identification problems be referred to your local or state Cooperative Extension Service agent. Color photographs and descriptions of important species can be found in Johnson and Lyon (1988). A detailed treatise on the biology, natural enemies, and control of aphids was edited by Minks and Harrewijn (1988).

IDENTIFICATION AND BIOLOGY OF APHIDS

Aphid identification

Other insects and abiotic disorders can produce plant injury similar in appearance to aphid injury. Use the information presented in Table 1 to distinguish between these.

Table 1. Distinguishing between aphids and aphid-like symptoms.

Problem	Symptoms	Time of Apperance	What to Look for
Aphids	New leaves are distorted	After flushes of new growth	Small (1/8") green, yellow, black, white, or orange insects are seen on new growth. White cast skins are seen. Honeydew and sooty mold are present Ref: Johnson and Lyon
2,4 D	All growth is	Often seen after windy or	No cast skins, honey dew or sooty mold

herbicide injury	distorted, twisted, including petioles and leaves	very hot weather. Appears very suddenly (24 hrs). Often only affects windward side of plant.	are present. Ref: Sinclair
Leafhoppers	New growth is stunted on leaf margins or interveinal areas are cholorotic or necrotic	Injury develops quickly (1 week)	Cast skins may be seen. Ref: Johnson and Lyon
Eriophyid mites	New growth, or current season's growth or buds are distorted, stunted and necrotic	Can be seen in spring or summer.	Very small, cigar-shaped mites with only four legs seen with a microscope. Ref: Johnson and Lyon

Family Aphididae

Aphids are small (usually less than 1/4" long), soft-bodied, pear-shaped insects. They may be pale yellow, green, red, blue, gray, or black, and may have spots or stripes. Winged forms have two membranous pairs of wings, with the front pair larger than the hind pair. Immature aphids closely resemble adults, but may differ in color and do not have wings. Most members of the family Aphididae possess a pair of elongate tubular structures, called cornicles, on the back of the fifth or sixth abdominal segment; in some species the cornicles are very small or absent. The antennae have six segments. Some species are covered by white, waxy fibers secreted from glands on the body, giving them a thick covering of fuzzy white wax; these are known as woolly aphids. Aphids are also characterized by the production of a sugary excretion called honeydew that may be produced in large quantities, often resulting in the growth of sooty mold on plant surfaces. This is unsightly and can reduce the photosynthetic capability of the plant.

Family Adelgidae

The adelgids have been called pine and spruce aphids in the older literature but are not true aphids. They lack cornicles and the antennae have three to five segments. All winged forms have five-segmented antennae, sexual forms have four-segmented antennae, and wingless parthenogenetic females have three-segmented antennae. Many species produce waxy threads that cover the body. Many species produce galls on spruce and some species, such as the balsam wooly adelgid, *Adelges piceae* (Ratzberg) and hemlock woolly adelgid, *A. tsugae* Annand, are capable of killing trees.

Family Phylloxeridae

The Phylloxeridae, or phylloxerans, are close relatives of adelgids and aphids. Like adelgids they also lack cornicles and in all forms the antennae are three-segmented. These insects do not produce waxy threads, but some species are covered with a waxy powder. Like some species of aphids and adelgids, many species of phylloxerins produce galls and some use multiple hosts.

Aphids, adelgids, and phylloxerans are distributed worldwide. Some species have restricted distributions that correspond to the range of their host plants. *Hamamelistes agrifoliae* Ferris is found only in California where it attacks coast live oak. Some species are more widespread; the woolly alder aphid, *Prociphilus tesselatus* Fitch, occurs in the east from Canada to Florida and west to the Mississippi River, and alternates between alder and silver maple in this range. A few species, including some of the most significant pest species, are cosmopolitan. The green peach aphid, *Myzus persicae* (Sulzer), is distributed worldwide.

Many pest species have been introduced from abroad. The balsam woolly adelgid, a native of Europe, was first discovered in North America in Maine in 1908 and quickly spread throughout the Appalachians from the maritime provinces of Canada to Georgia and North Carolina. In 1928 it was discovered in the Pacific Northwest and now extends along the Coast and Cascade mountains from British Columbia to California. The hemlock woolly adelgid, *Adelges tsugae* Annand, is probably a native of the orient. It was first reported in this country in 1927 and now occurs in California, the Pacific Northwest, and from the maritime provinces to the mid-Atlantic states in the east.

Aphids consume only plant material and are most often found on their host plants. Different species show a preference for different hosts, and their habitat is in large measure determined by the distribution of their hosts. Species habitats are further defined by their location on the host plant. Many species are foliage feeders, others prefer tender shoots and stems, some are found on the woody parts of trees and shrubs, and a few are found on the roots. Some species utilize different parts of the host at different times of the year, their choice being dependent on the season, stage of growth, or the species of host plant. Each host plant has its own chemistry, so aphids may select one individual plant in a stand and feed on it exclusively.

Wind may dislodge aphids from their host plant by causing the leaves to rub together, knocking off the insects. Wind also is considered the most important factor in the dispersal of aphids from one plant to the next, as in the case of the balsam woolly adelgid, and in the migration of winged forms from one area to another. Because of their small size and weak flight muscles aphids, adelgids, and phylloxerans are not strong fliers and have little control over the direction of their flight when the wind speed is more than a few miles per hour.

Aphids often have very complicated life cycles that involve alternation of host plants, sexual and asexual generations, and winged migrant and wingless nonmigratory generations. In temperate regions most species overwinter as eggs, and the eggs hatch in the spring into females that give birth to live young. Several generations may be produced asexually, resulting in very rapid growth of populations. In some species, such as the apple aphid, *Aphis pomi* De Geer, the aphids may remain on a single host throughout the year. In other species, winged females migrate to new plants and produce more young asexually. These hosts may be quite different from the primary host. For example, the rosy apple aphid, *Dysaphis plantaginae* (Passerini), moves from a woody primary host (*Malus*) to a herbaceous secondary host (*Plantago*). Late in the season, a sexual generation consisting of winged males and females is produced. These return to primary hosts where they mate and the females lay eggs that overwinter. Details of the life cycle and patterns of host utilization vary considerably among aphid species.

Adelgids also can have complex life cycles that include sexual and asexual generations and alternation of hosts. Many species of adelgids utilize spruce as a primary host and other conifers such as pines, larch, firs, and Douglas fir as secondary hosts. For example, the Cooley spruce gall adelgid causes pineapple-like galls on spruce and needle distortion on Douglas fir. Another unusual life cycle is that of the balsam woolly adelgid. Populations in North America consist entirely of females. They do not give birth to live young, but instead lay eggs. There are two to four generations per year, depending on the locality and elevation of the population, with fewer generations produced in the northern parts and higher elevations within the range.

Phylloxerans produce galls on several species of deciduous plants. In most species of phylloxerans the entire life cycle is completed on a single host. However, in at least two species, *Phylloxera texana* Stoetzel and *P. castanea* Pergande, *Carya* serves as the primary host and oak or hickory are secondary hosts (Stoetzel 1985).

The level of precipitation also affects the vigor of the host plant, which affects the aphids feeding on it. Some aphid species do better when a plant is well-watered and fertilized, while others do better if the plant suffers from stress.

The development of winged individuals in a population seems to be triggered by the degree of crowding on the host plant, but the way in which this works depends on the species (Hille Ris Lambers 1966). Temperature, daylength, and host plant food quality all combine to play a role in aphid dispersal.

Ants have an important role in the development and success of many aphid species (Way 1963). The presence of aphid-tending ants may inhibit the production of winged forms. The ants collect honeydew excreted by the aphids and stimulate the aphids to produce large quantities of honeydew by stroking them with their antennae. Ants protect aphids from parasites and predators, and even transport them to suitable host plants and safe places to hibernate. Some species of aphids live in the nests of their benefactors and are dependent on their ant protectors for their survival. Monitors should look for ants as a clue to the presence of aphids.

Certain horticultural practices may affect the abundance of aphids and adelgids. It has long been known that plants and plant parts rich in nitrogen are exploited by aphids (Minks and Harrewijn 1987). Recently, McClure (1991) demonstrated that populations of hemlock woolly adelgid increased dramatically when hemlocks were fertilized. Resource managers should be aware that the over- fertilization of plants may facilitate population increases of some associated pests such as aphids and adelgids.

Feeding by aphids, adelgids, and phylloxerans withdraws sap from the host plant and can interfere with the physiology of the plant by altering the balance of plant growth hormones. Aphids feeding on leaves can cause yellowing, spotting, and premature death, and can reduce the ability of the leaf to photosynthesize by reducing the amount of fluid in the leaf and reducing the surface area as a result of curling. Aphids are believed to inject toxins into plant tissues as they feed. These toxins may produce local and systemic effects in plants that include reductions in growth and alterations in the normal patterns of nutrient distribution in the plant (Minter and Harrewijn 1987). Twigs may develop swelling or gouting.

The production of large amounts of honeydew by aphids may foul plant tissues and structures, walkways, or vehicles beneath heavily infested trees. The honeydew may attract stinging insects, ants, and other insects and may create a nuisance. Furthermore, honeydew on plant tissues may facilitate the growth of sooty mold which itself is unsightly and may reduce the photosynthetic capacity of plants.

Many species of aphids, adelgids, and phylloxerans produce galls on their host plant. Galls are distinctive growths on leaves, shoots, stems, or roots, that are a response of the plant to certain stimuli provided by the aphids. Galls such as those produced by adelgids on spruce may interfere with normal patterns of plant growth by killing terminals. Leaves heavily galled by phylloxerans may be dropped prematurely. Galls may also reduce the aesthetic quality of plants. In some cases it is possible to identify the species of aphid appearance of the gall. Useful references include Felt (1940), Johnson and Lyon (1988), and Russo (1979).

Aphids and adelgids can cause distortions of other plant parts. Symptoms caused by balsam woolly adelgid on fir begin with curling and dieback of the current year's growth, swelling of buds and gouting of shoots, and thinning of the crown. In trees with a heavy infestation on the bole, the wood becomes reddish and coarse, a condition known as "rotholz" or redwood (Knight and Heikkenen 1980). Susceptibility varies with the species; subalpine fir dies within a few years, sometimes before terminal swelling occurs; Grand fir may survive fifteen years; Noble fir, Shasta red fir, and white fir may show gouting but usually are not killed. In Great Smoky Mountains National Park, balsam woolly adelgids kill Fraser fir in two to six years (Allen-Reid 1984).

In addition to direct injury caused by their feeding, aphids are serious plant pests because of their role as vectors of plant diseases. Hundreds of plant viruses are transmitted by aphids. The green peach aphid, *Myzus persicae* (Sulzer), is the single most important vector. It is known to transmit over 100 virus diseases to plants in about 30 different families (Ossiannilsson 1966; van Emden et. al. 1969). The ability of aphids to transmit plant diseases is related to their piercing-sucking feeding habit, rapid growth, and life histories that involve host alternation and migration.

MONITORING AND THRESHOLDS FOR APHIDS

Population Monitoring Techniques for Aphids

At least two methods have been used for monitoring aphids, adelgids, and phylloxerans. The first and most commonly used technique involves visual observation of the plant. On small plants, examine the entire plant; on larger plants and trees, examine representative leaves, twigs, stems, or other portions of the plant. The part of the plant to be examined also will be determined by the biology of the pest. For example, in the Great Smoky Mountains National Park, balsam woolly adelgid infestations are greatest at about 15' above the ground rather than at breast height, the standard position for sampling on trees. Therefore, monitoring populations of this pest is done at this greater height (C. Eagar, pers. comm.). Data can be recorded on monitoring forms such as the one shown in Table 2.

A system of classifying relative levels of infestation was presented by Heathcote (1972). He used the following population density index to classify infestations.

None (O) - no aphids seen.

Very light (V) - one to a few aphids per plant and only a few scattered young plants infested, or one to a few aphids per leaf, shoot, or other section of larger plant or tree and only a few colonies per large plant with the colonies on the young tender leaves or buds.

Light (L) - 5-25 aphids per plant and many plants infested, or with many colonies on larger plants or trees, and the colonies not confined to young shoots.

Medium (M) - 25-100 aphids per plant and most plants infested, or with large numbers of aphids on larger plants or trees and not in recognizable colonies, but diffuse and infesting many leaves, stems, etc.

Heavy (H) - more than 100 aphids per plant with virtually all plants infested, or with stems, leaves, buds, etc., solidly covered with aphids.

Where direct observation of aphids is difficult, such as in tall trees, monitoring may be done indirectly by quantifying production of honeydew. Dreistadt and Dahlsten (1988) used water-sensitive spray droplet cards to collect honeydew beneath tall tuliptrees infested with aphids. The honeydew droplet counts correlated well with the abundance of aphids in the canopy of the trees.

When monitoring through visual observation, also survey for the presence and effectiveness of aphid pathogens, parasites, and predators. Aphids killed by fungi, bacteria, or other pathogens usually remain on the plant and can be recognized by their immobility and peculiar coloration. Parasitized aphids usually are darker than unparasitized aphids, at least near the completion of the parasite's life cycle. Aphids that have been killed by parasitic wasps are mummified; that is, they are discolored and papery in texture. If the adult wasp has emerged there is a round hole in the mummy where the wasp exited. Color photographs of aphid mummies are given in Yepsen (1984). Look for predators among the aphids on the plant, as well as flying or perching nearby.

Decision-Making and Thresholds for Aphids

Data on the number of aphids, adelgids, and phylloxerans may be combined with other information, including the injury caused by the pest, the value of the plants being managed, and the cost of control activities, to create economic or aesthetic injury levels and thresholds (Raupp et al. 1988, 1989). Because of the economic importance of aphids, thresholds and action levels have been established for several aphid-crop systems. However, there has been relatively little work concerning aphids, adelgids, and phylloxerans infesting ornamental plants. Notable exceptions include the following studies. In 1978 Olkowski et al. published a decision-making guideline for ornamental spruces being attacked by the blue spruce aphid, *Elatobium abietinum* Walker. They determined that about 34 aphids per quadrant of a tree collected with a beating cloth were sufficient to cause defoliation. They used this level as an aesthetic injury level to

justify intervention. In 1988 Dreistadt and Dahlsten presented a methodology for determining a threshold for managing the tuliptree aphid, *Illinoia liriodendri* (Monell), based on honeydew counts and public complaints about the honeydew. This methodology could be used as a model for establishing decision-making rules for other aphid problems in the National Park Service. Finally, Nielsen (1989)
published action thresholds for aphids found on the leaves of hardwood trees. He suggested that two aphids per leaf in the spring and four aphids per leaf in the summer justify intervention.

In setting thresholds and action levels in the National Park Service, the particular needs of each park must be considered. Unless threatened or endangered plants are being attacked, control is not generally recommended in natural areas. Under normal circumstances natural mortality factors will keep aphid populations in check in these areas. However, this may not be the case for certain exotic species such as balsam and hemlock woolly adelgids. These species appear to lack natural enemies in this country that are capable of establishing control.

Thresholds will be quite low and may approach zero for plants that are valuable due to their size, age, beauty, rarity, or historic significance. Low thresholds will also be established for plants that are extremely vulnerable to a pest capable of causing death, such as Canadian hemlocks under attack by the hemlock woolly adelgid. Specimen plants and small groups of plantings are good candidates for the establishment of thresholds and other decision- making guidelines.

A further complication arises if aphid-borne diseases threaten plants. If this is the case, the threshold level will be much lower than for aphid damage alone. Accurate identification of both the aphid vector and the disease is essential to be positive that the suspected vector and the disease are causally related. Consult diagnostic experts at your Cooperative Extension Service or commercial laboratory for aid in identification of aphids and plant diseases that may be transmitted by them.

NON-CHEMICAL CONTROL OF APHIDS

1. Avoid planting species and cultivars susceptible to aphids, adelgids, and phylloxerans.

2. Begin monitoring plants early in the growing season and record your observations using a standardized system such as the one presented in the decision-making section. Record the presence of aphid parasitoids and predators.

3. If decision-making guidelines have not been established for the resource under management, use the methodology outlined in the decision-making section to establish thresholds or action levels. Intervention may also be necessary if sooty mold or honeydew are problems.

4. If aphid populations exceeded thresholds in the previous season, consider using a dormant oil to kill overwintering life stages.

5. Use mechanical and cultural controls where feasible. Use aluminum foil or white plastic mulches in newly planted areas if possible. The reflection of light from these materials will

confuse aphids and prevent them from landing on plants. Crush aphids with fingers if infestations are not too extensive. Control aphid-tending ants by preventing them from reaching plants, using sticky substances around tree trunks, or bone meal or crushed charcoal barriers in and around gardens. Destroy ant colonies if necessary. Eliminate alternate host plants in the vicinity of more desirable plants.

6. Encourage natural predators and parasitoids. Release lacewings, ladybird beetles, syrphid flies, predaceous midges, and aphid mummies. Plant nectar-producing flowering plants that attract adults of these insects. Provide suitable habitat that will encourage predators to remain in the vicinity.

7. Consider release of exotic parasitoids and predators in cooperation with federal and state experts.

8. Spot treat with insecticidal soap, oil, or other approved insecticides when necessary. Spot treatment will have less impact on biological control agents than widespread spraying.

Biological Control

Aphids, adelgids, and phylloxerans have many natural enemies, including diseases, parasites, and predators. The ecology of aphid predators has been reviewed by Hodek (1966), and the impact of the natural enemies of aphids has been reviewed by Hagen and van den Bosch (1968). Reviews dealing with specific groups of pathogens, parasites, and predators of aphids include Madelin (1966), Hodek (1967), Schneider (1969), Stary (1970), Ferron (1978), Hall (1981), Wilding (1981), Viggiani (1984), and Minks and Harrewijn (1988).

Diseases of aphids include several species of fungi that are capable of drastically reducing aphid populations under appropriate conditions. Excessive moisture in cool weather favors the development of entomogenous fungi. Outbreaks of fungal pathogens are more likely to occur in cool, moist seasons than in warm dry seasons. Aphids also are susceptible to infection by bacteria, viruses, protozoa, and nematodes, but none of these is known to cause high mortality in natural populations. Pathogens for the control of aphids have been used successfully in greenhouses but only with limited success in field situations. A fungal pathogen, *Verticillium leucanii*, is available commercially in Europe, but not yet in the United States.

Aphids are parasitized by many insects, the most important belonging to the hymenopteran families Aphidiidae and Aphelinidae. The family Aphidiidae contains over 300 species, all of which are parasites of aphids. In the family Aphelinidae, only species in the genera *Aphelinus*, *Mesidia*, and *Mesidiopsis* parasitize aphids, but certain of these have proven successful in biological control programs. Two other hymenopteran families, the Encyrtidae and the Mymaridae, also include aphid parasites. Similarly, many insects feed on aphids, including beetles, flies, lacewings, earwigs, and predaceous bugs. Ladybird beetles (Coccinellidae) and green lacewings (Chrysopidae) feed on aphids as larvae and adults. Hover flies (Syrphidae) and predaceous gall midges (Cecidomyiidae) eat aphids only as larvae. All are thought to play important roles in reducing aphid populations (Hagen and van den Bosch 1968, Minks and Harrewijn 1988). Few vertebrates have been reported feeding on aphids, but Smith (1966) reported that in Great Britain birds may have a significant impact on aphid populations under some circumstances.

Many parasites have been successfully introduced for control of aphids (Clausen 1978, Olkowski et. al. 1976, Minter and Harrewijn 1988). Parasites often are specific for one or a few aphid species, requiring accurate identification of the aphid species in order to match the correct parasite species to the problem. Consult with federal and state extension officials and your regional National Park Service Integrated Pest Management coordinator before considering implementation of a parasite release program. Predators also have been used successfully against some species of aphids. Specific predators have been imported from overseas to help combat a variety of aphids (Mitchell et al. 1970). Native predators, such as ladybird beetles, lacewings, predaceous midges, and syrphid flies have also given good results. Ladybird beetles, lacewings, midges, and several species of parasitoids can be obtained from commercial supply houses. A list of suppliers is available through the Biological Control Services Program, 3288 Meadowview Road, Sacramento, CA, 95832.

Natural predators and parasites may be augmented by various techniques. A sugar or sugar and protein food supplement may be sprayed on plants to attract green lacewings and ladybird beetles (Hagen et. al. 1970; Schiefelbein and Chiang 1966). Commercial preparations of such supplements are available. Larvae of predators and aphid mummies may be collected in one area and released in the control area. The adults of predators such as syrphid flies and parasitic wasps may be encouraged to stay in an area by planting nectar producing flowering plants to provide food for the adults. Carroll and Hoyt (1984) report good control of apple aphids in orchards by using earwigs reared on dog food and released at five or six per tree. "Earwig retreats" made of cardboard and paper towels were placed in the trees and straw was scattered on the ground under the trees to encourage the earwigs to stay. Aphid densities declined dramatically in augmented trees.

Beneficial organisms may also be preserved by using pesticides with short residual activities such as soap and oil and avoiding treatments of large numbers of plants in favor of spot treating only individual plants that require intervention.

Resistant Varieties

Use of plant cultivars and species that are less susceptible to these aphids should be encouraged. Sadof and Raupp (1991) reported that aphid populations increased more on variegated euonymus cultivars than on green cultivars. Cranshaw (1989) found that green individuals of Colorado blue spruce were more likely to be infested by the Cooley spruce gall adelgid compared to blue individuals. Avoid planting Douglas fir near spruces infested with Cooley spruce gall adelgid as Douglas fir is the alternate host for this species.

Physical Control

A simple approach to aphid control is to crush them between your fingers. This will work on garden plants and other ornamentals with light infestations. Another simple approach is to knock the aphids off the plants with a stream of water from a hose or sprayer, although the efficacy of this technique is unknown.

Cultural Control

Cultural practices may also reduce aphid problems. Aluminum foil and white plastic mulches can inhibit the migration of winged aphids into newly planted areas (Wyman et. al. 1979, Yepsen 1984). These work best with young, small plants up to 1' tall. The highly reflective surface of the mulch causes migrating aphids to become disoriented, reducing the number of migrants that land and become established on the plants. Controlling alternate hosts of the pest species can also successfully control aphids (Knipling 1979). For example, to control the green peach aphid in gardens and orchards, Yepsen (1984) recommends clearing plants such as plantain, bindweed, and lamb's quarters from nearby land.

Ants play an important role in the success of aphids. Therefore, control of ant populations can cause a significant reduction in aphid populations. If ants are observed on aphid- infested trees, apply a commercial adhesive designed for such purposes in a band around the lower trunk of the tree. Caution should be exercised in applying these materials directly to the bark of trees; they can cause long-lasting discoloration or may injure thin-barked trees. In situations where individual treatment of plants is impractical, a barrier of bone meal or crushed charcoal may keep ants away. Destroy colonies of aphid-tending ants, if necessary; keep in mind that ants often are beneficial insects and eliminating them may not be the best strategy.

CHEMICAL CONTROL OF APHIDS

Dormant oils are applied during the dormant season of the plant, either in fall or spring before bud break, to kill eggs or other overwintering life stages on oil-tolerant plants. Recent improvements in the formulation of oil products have facilitated the use of these materials on a wide variety of plants during the growing season. Used in the nondormant seasons, summer oil or horticultural spray oil, has proven very effective in reducing population of adelgids on ornamental trees (McClure 1987, Baxendale and Johnson 1990). The efficacy of oils in controlling aphids has been equivocal. For some species such as the crapemyrtle aphid, *Tinocallis kahawaluokalani* (Kirkaldy), oil provided good control (Booth et al. 1990); for other species such as the birch aphid, *Euceraphis betulae* (Koch), oil provided little or no control (Nielsen 1990).

Insecticidal soaps are also recommended for aphid control. Like oils they have been effectively used to control adelgids (McClure 1987). However, their efficacy against aphids varies (Booth et al. 1990, Nielsen 1990).

Several conventional pesticides control aphids. Contact your regional Integrated Pest Management coordinator to determine which, if any, pesticide is best suited for your management program.

APHID INTEGRATED PEST MANAGEMENT DEMONSTRATION PROGRAMS

In an integrated control program in several cities in California, many aphid management techniques were combined to provide superior levels of aphid control and a dramatic reduction in pesticide use (Olkowski 1973, Olkowski et al. 1976, Flint and van den Bosch 1981). The first step was to institute a monitoring program to accurately assess the aphid problem on trees lining city streets. Pest species were identified. Aesthetic injury levels were established and management techniques were applied only if the thresholds were exceeded. Parasites of exotic aphid species were located and imported for release. Several species of imported parasites became established and have contributed to the management program. Heavily infested trees were pruned to remove the highly susceptible inner canopy. Where aphid-tending ants interfered with predators and parasites, bands of a commercial sticky substance were applied around the bases of trees. In Berkeley, where the program began, pesticide usage went from hundreds of pounds per year to zero, and the aphid problem became negligible.

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PERSONAL COMMUNICATIONS

Christopher Eagar
Biological Technician
Uplands Field Research Laboratory
Great Smoky Mountains National Park
Gatlinburg, Tennessee 37738

Manya Stoetzel
Research Entomologist
Systematic Entomology Laboratory
USDA, ARS
Beltsville, Maryland 20705

Dutch Elm Disease

This module is intended to serve as a source of basic information needed to implement an Integrated Pest Management program for Dutch elm disease. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

IDENTIFICATION AND BIOLOGY OF DUTCH ELM DISEASE

Is it Dutch Elm Disease?

An IPM program for elms is much more than managing a disease (Dutch elm disease) and its insect vectors (european elm bark beetle and native elm bark beetle). Not only are there several alternatives available for the management of these pests, there are also several other pest problems of elms which can cause symptoms similar to Dutch elm disease; it is essential that the tree manager be familiar with these as well. Insect and disease problems which cause symptoms similar to Dutch elm disease are summarized in Table 1.

Table 1. Diagnosis of elm disorders.

Symptom	Time of appearance	Possible causes	Diagnosis
Failure of new growth to develop in spring	March-April	DED, scale insects, limb injury.	DED—examine wood for streaking, culture for disease organisms. Look for evidence of beetle feeding in branch crotches. Scale—look under covers and remove to determine viability Ref: 6,13,17,18
New growth develops poorly in the spring (slow leaf expansion and shoot elongation)	March-April-May	DED scale insects, nutrient deficiency, elm yellows.	DED, scale—see above. Have soil and tissue nutrient tests performed for deficiency, nutrient levels. Determine elm fertilization history of site. 'Phloem necrosis'-sudden necrosis and death of undeveloped leaves. Streaking in phloem tissue accompanied by wintergreen odor. Ref: 6, 9, 13, 17, 18
Sudden yellowing and necrosis on a branch or branches	Anytime during growing season	DED, elm See above.	
Uneven browning of leaf margins on a section of the tree	July-end of scorch season	Biotic leaf scorch, branch or root system injury, chemical phytotoxicity.	Biotic – positive culture for bacterial branch or leaf scorch. Trace injury or phytotoxicity to chemical application. Ref: 8, 13, 17, 18

Uniform browning of leaf margins	May-end of growing season	Abiotic leaf scorch, root system injury, chemical phytotoxicity	Abiotic—negative culture for bacterial leaf scorch. Hot, dry, weather prior to development of symptoms.Trace injury to phytotoxicity to chemical application. Ref: 8, 13, 17, 18
Skeletonization or small holes in leaves	May, July	Elm leaf beetle larvae	Small holes in leaves from feeding by adults, skeletonization from Elm leaf beetle larvae. Both are ¼” to 3/8” in length and yellow with black stripes. Ref: 6, 7
Brown spots along leaf veins	Late April-Late May	Anthracnose	Results after a cool, wet spring. Ref: 17, 18

Dutch Elm Disease and Its Insect Vectors

Dutch elm disease is caused by the fungus *Ophiostoma ulmi* (Buism.) Nannf. (=*Ceratocystis ulmi* (Buism.) C. Moreau). (For a complete description of the life cycle of the pathogen, see Sinclair et al. 1987.) *O. ulmi* is an introduced pathogen that arrived in North America in elm logs from Europe. The pathogen overwinters in the bark of infected trees or logs cut from infected trees. It is carried from infected to uninfected trees by two insect vectors, the European elm bark beetle (*Scolytus multistriatus*) and the native elm bark beetle (*Hylurgopinus rufipes*). Althogh the European elm bark beetle is the major vector of the disease, temperatures below -6F kill the larvae; thus the native elm bark beetle is the primary vector in parts of the northern United States, New England, and all of Canada. Both species of beetles bore into the bark of infected trees and excavate egg galleries. The galleries of *S. multistriatus* run parallel to the grain of the wood, while those of *H. rufipes* are at a 45 degree angle to the grain. Larvae hatch from the eggs in approximately one week; the white, legless grubs tunnel perpendicularly to the maternal gallery, feeding on elm phloem cells for 4 to 5 weeks. A 1 to 2 week long pupal stage follows. The 1/8" adult beetles tunnel to the bark surface to emerge and fly to new trees. As they move to the surface, fungal spores that have germinated and spread throughout the feeding galleries attach to the beetles' bodies. These spores are then carried to new trees as the beetles move there to feed. The beetles can fly for several miles, allowing the disease to spread over a wide area. The pathogen also moves between trees via root grafts. The fungal spores move passively within the tree in xylem vessels (both up and down the tree from the point of infection). The fungus also moves actively between xylem vessels as fungal hyphae. The pathogen kills the tree by blocking solute movement in the xylem as well and by producing a toxin. Acute and chronic forms of the disease are recognized. The acute form is thought to cause wilt and branch death while the chronic form is thought to lead to more gradual chlorosis and leaf drop.

After emergence from the brood tree, adult beetles fly to other elms to feed and to breed. Bark beetles can also feed on logs cut for lumber and fuel. Such colonized wood may become a reservoir for beetles and hence Dutch elm disease even if the wood is of a resistant elm species. This is why debarking of elm wood that is being stored for use as fuel is stressed as part of the management strategy for control of Dutch elm disease.

S. multistriatus commonly produces two generations per year; the first overwinters as larvae, pupates in early spring, and emerges as elms reach full leaf. The second generation flies in late summer, producing the broods that overwinter. *H. rufipes* usually overwinters as adults in the bark at the base of healthy elms, and may infect the tree with Dutch elm disease during fall feeding in lower boles or spring feeding in branches. *H. rufipes* may produce one to one and one-half generations per year, overwintering as either larvae or adults.

Pheromone traps for elm bark beetles are available and can be used to monitor beetle populations. Information on flight activity is useful if insecticides are to be used to control adult beetles.

The Dutch elm disease pathogen, *O. ulmi*, grows and sporulates in elm tissues throughout the growing season. The sporulation of the fungus is temperature-related. Asexual spores are most commonly produced during the warm months. When sexual reproduction takes place (which is rare in nature) the production of fruiting bodies increases, occurring most commonly between November and February.

Diagnosis of Dutch Elm Disease by Testing for the Pathogen

Pathologists now recognize that there are several strains of Dutch elm disease, and that some are more aggressive than others. These strains kill elms more rapidly (Richards and Takai 1984). The production of toxins by the pathogen was first recognized in 1947 (Diamond 1947), and aggressive and non-aggressive strains were first recognized in 1984 (Sinclair et al. 1987). More recently, an antibody specific for some of the Dutch elm disease toxins was produced (Benhamou et al. 1985). This will enable the development of a fast, accurate test in which fluids from the suspect tree are matched against Dutch elm disease toxins. The nature of the reaction would confirm or deny the presence of the disease. Current tests for the pathogen involve culture of the disease organism from diseased wood, a procedure which can take several days. This test is usually recommended only in areas where the disease has not been previously reported.

NON-CHEMICAL CONTROL OF DUTCH ELM DISEASE

Prior to the introduction of Dutch elm disease into the United States in 1930, the American Elm (*U. americana* L.) was one of the most popular street trees throughout the Northeast, Middle Atlantic, and Midwest; about 77 million elms were growing in the United States. By 1976, about 43 million of those trees had been lost to Dutch elm disease (USFS 1977). The impressive shape, size, fall color, and shade quality of the American elm led to the institution of a near monoculture of this species in many urban areas, which has served to enhance the spread of Dutch elm disease. Maintenance costs for Dutch elm disease management vary considerably depending on the community and the nature of its elm plantings; they were estimated to range between $26,000 and $152,000 (in 1982 dollars) annually in several communities in a four-year demonstration project (Hanisch et al. 1983).

A management program for Dutch elm disease encompasses many different strategies. Factors to consider in deciding which are most appropriate for a situation include the time of year, the resources at your disposal, and the number and location of trees affected.

Resources available for Dutch elm disease management are often limited, so the tree manager must often balance the value of the trees in the landscape and the degree of infection in making a decision about managing Dutch elm disease. From an aesthetic point of view, trees in natural areas are generally considered least important when funding for maintenance is limited. The drawback to this approach is that these trees may be close enough to valuable plantings to form root grafts or to serve as a breeding ground for beetles which can then fly to those plantings and infect them with the disease. In a study on the use of pheromone traps for mass trapping, beetles were caught as far as five miles from the nearest elm tree (Birch et al. 1981).

The total amount of tree biomass affected must also be considered. Several authorities on Dutch elm disease feel that it is most realistic to expect to control a small infection on a large tree (Lanier 1988). Source of infection is also important; as presented in Table 2, trees with root graft infections were not successfully treated by any available method; trees with current season's infections were treated with a higher success rate than those with residual infections. It was also suggested that fungicide injections directly into branches with localized infections rather than into the bole of the tree (the current practice) would greatly increase the success rate of therapeutic injections (Lanier 1988).

Use of Resistant Varieties

The severity of Dutch elm disease and the desirable aesthetic qualities of the elm have led to the development of several elm varieties with resistance to Dutch elm disease as well as re- planting with tree species that have some of the ornamental characteristics of the elm. American, English, red, and winged elm are among the most susceptible, while Chinese, Japanese, and Siberian elms are among the most resistant. Resistance is not the same as immunity, however. The presence of Dutch elm disease was recently confirmed in several Chinese elms planted adjacent to the National Mall in Washington, D.C. Several resistant cultivars have been developed from crosses of European and Asian elms. While many of these trees are promising, none seems to be as attractive as the American elm. They also have not been planted long enough for all their possible insect, disease, and cultural problems to have been recognized. For example, the Siberian elm is highly susceptible to the elm leaf beetle, as is the Japanese zelkova. (See Sinclair et al. 1987 for more information.) Other disorders, such as bacterial leaf scorch and elm yellows must also be considered. The mechanism of resistance seems to be related to the ability of the tree to quickly heal the wounded area and thus prevent the movement of the pathogen to other parts of the tree.

Sanitation

This is the most important element of a Dutch elm disease management program for existing elms because it removes the elm bark beetle's breeding habitat from the system. No Dutch elm disease management program will be successful without good sanitation. It consists of the immediate removal of any dead or wounded branches, and the debarking of branches stored for use as firewood. Flagging branches on which streaking has been observed are also removed. Ideally, branches should be cut back 10' from the last point where streaking is evident. This is determined by making small cuts in the bark to look for streaking. The final pruning cut for removal of the branch should be made approximately 10' behind the point at which healthy wood is first observed (Lanier 1988).

Sanitation should be viewed as a community-wide management tactic. Considering the distance that elm bark beetles can travel, removal of branches from a single tree will have little impact in the infection status of that tree if there are other infected trees in the area. Sanitation, while a key component of a Dutch elm disease management program, is most effective when combined with the judicious use of fungicides, as outlined by Lanier (Lanier 1988). This is discussed in more detail in the section on chemical control.

Pruning Schedules

Wounding trees by pruning will attract the bark beetle vectors of Dutch elm disease (Byers et al., 1980). Ideally, routine pruning should be done in the dormant season. If this is not possible, pruning of healthy elms should be restricted to periods of beetle inactivity. This can be determined by the use of pheromone traps to monitor beetle flight periods.

Mass-trapping of Elm Bark Beetles

Mass-trapping of beetles using pheromone traps has also been investigated as a way to control the European elm bark beetle and thus reduce the spread of Dutch elm disease. A study in California estimated that only 1%-20% of marked beetles released in three study areas were recaptured, indicating that traps alone are not sufficient to reduce beetle populations (Birch et al., 1981).

CHEMICAL CONTROL OF DUTCH ELM DISEASE

Lanier (1988) suggests that pruning combined with fungicide gives better disease management than pruning or fungicides alone when dealing with a residual infection. Fungicides were most effective when injected directly into large limbs where an infection had been found, as well as into the bole. For current year infections, pruning alone was as effective as pruning with the use of a fungicide. These results are summarized in Table 2.

Table 2. Effectiveness of Dutch elm disease management strategies based on history of the infection (based on Lanier 1988).

Infection History	Management Strategy	Effectiveness
Current year	Pruning	100%
	Injection	76%
	Both	100%
Residual	Pruning	0%
	Injection	33%
	Both	71%
Root Graft	Injection	0%

Infection history can be determined by noting when and where on the tree the symptoms develop. Symptoms that appear during the first eight weeks after leaf development are most likely the result of a residual infection or a root graft, while those that appear later than this result from a current season's infection. Symptoms appearing on several branches at the same time suggest that either a root graft or multiple beetle infections. If there is a multiple-branch infection on a limited section of the tree, suspect disease transmission via a root graft.

As shown in Table 2, the effectiveness of fungicide injections varies considerably depending on the nature of the Dutch elm disease infection source. Recommended timing of sprays to control elm bark beetle varies as well; both winter (Hanisch et al., 1983) and early spring (Davidson 1991) treatments are recommended. Only the latter corresponds to a point in the beetle's life cycle when effective control can be obtained. See Birch et al.(1981); Johnson and Lyon (1988); Pajares and Lanier (1989) for more information on choice and timing of insecticides for beetle control.

Trap-tree Strategy for Dutch Elm Disease Management

The use of trap trees is an alternative type of sanitation for Dutch elm disease management. In this approach, most recently reviewed by Lanier (1989), trees infected with Dutch elm disease that cannot immediately be removed are injected with the herbicide cacodylic acid which causes the tree to die quickly. The goals of this approach are to remove infected trees from the system when the labor or money to take them down is not available and to attract beetles away from healthier trees to the dying tree. Beetles oviposit in the dying tree but the larvae do not survive because the tree is dead when the eggs hatch. Cacodylic acid has been used successfully in parks where site factors made tree removal impossible. This strategy is affected by the following conditions (Lanier 1989; Lanier and Jones 1985; O'Callaghan et al. 1980).

1) The amount of beetle mortality is affected by the degree of dieback in the crown at the time of cacodylic acid application. There will be much greater mortality if less than 50% of the crown is dead than if more than 90% is dead.

2) If possible, time cacodylic acid applications to periods just before adult flight begins.

3) Herbicide will move through root grafts, so trees to be treated must also be trenched.

4) For maximum beetle mortality, bait the tree with a pheromone trap and treat the lower bole with an insecticide. Lanier and Jones (1985) found that addition of an insecticide to this system increased beetle mortality. For every beetle which landed on and bored into a tree treated with cacodylic acid, 40 landed and did not bore into the tree. Thus the use of an insecticide greatly enhanced beetle mortality.

MANAGEMENT OF OTHER ELM PESTS

ELM YELLOWS

IDENTIFICATION AND BIOLOGY

Elm yellows (also known as elm phloem necrosis) is caused by an organism called a "mycoplasma-like organism" (MLO) which biologists classify between a virus and a bacterium. It is carried between elm trees by leafhoppers and possibly spittlebugs (Sinclair and Johnson 1987). Roots are infected first, where root tips and root hairs are killed. Foliar symptoms appear in mid-summer of the second season of infection. Leaves droop and curl, turn yellow and then brown. The disease is reported to be fatal in elm species native to North America but not in elms of European or Asiatic descent. Death is sudden, occurring within a few weeks after the onset of foliar symptoms.

MLOs cannot be cultured in the laboratory, so diagnosis is performed in the field on living but infected trees. When the bark is peeled back it is be yellow or tan, sometimes speckled with brown (the normal bark color is creamy white). Infected bark also emits a strong wintergreen odor. If this is not noticed at first, warming a bark sample in a pocket will enhance the odor (Holmes 1987).

CONTROL OF ELM YELLOWS

The management strategy for this disease is simple. Dead trees **must** be removed **immediately**, and control measures must be applied for leafhoppers. Timing of leafhopper control coincides with the two flushes of elm shoot growth (Holmes 1987). While the disease has been reported throughout the eastern United States and southern Ontario (Sinclair et al., 1987), its occurrence is sporadic; thus control of the vectors is recommended only when the disease is known to be in the area.

SCALE INSECTS

IDENTIFICATION AND BIOLOGY

The European elm scale (*Gossyparia spuria*) as well as several species of leucanium scales produce symptoms similar to Dutch elm disease on elm. For more information on the biology and identification of scale insects that feed on elm, consult Johnson and Lyon (1988). The important point is that dieback and poor growth, which are symptoms of Dutch elm disease could also be caused by scale organisms, so this must be ruled out (or the presence of Dutch elm disease confirmed) before a tree is treated for Dutch elm disease.

CONTROL OF SCALE INSECTS

Management of scale insects is straight-forward. Once the presence of scale has been determined, remove several scale covers to confirm that the scales are viable. If this is so, apply horticultural oil either during the dormant season or during the growing season when scale crawlers are active. The timing of the latter spray will depend on the specific scale insect and geographic location of the trees. A dormant oil application in late March would be the most effective treatment since there is no leaf surface to intercept the spray. An application in March will have some impact on the elm bark beetles that will have started to feed.

ELM LEAF BEETLE

IDENTIFICATION AND BIOLOGY

The elm leaf beetle (*Pyrrhalta luteola*) is a common defoliator of all elm species as well as the Japanese zelkova, a frequent elm replacement. It overwinters as an adult in buildings or protected outdoor sites. It lays eggs on the expanding leaves in the spring. Females feed on elm leaves before ovipositing. The larvae feed by skeletonizing the leaves, which causes the leaves to appear yellow or brown from a distance; thus their injury might be mistaken for Dutch elm disease at first. The beetle larvae move to the base of the tree to pupate; adults emerge one to two weeks later. The pupae are bright yellow and may be seen in masses on the soil at the base of a tree. There are one to three or more generations per year depending on the latitude.

Leaf beetle larval activity can be monitored by the use of tree bands. Place a band coated with sticky material around the tree; as the beetles move up or down they will be caught. A large increase in larval catch indicates they are moving to the tree base to pupate. The larvae are about 3/8" long, and are yellow with black stripes running the length of their bodies.

CONTROL OF ELM LEAF BEETLE

The beetle can be controlled with a strain of *Bacillus thuringiensis* and several conventional pesticides, some of which may be applied to the bark of the tree to kill migrating larvae. In the case of a small infestation on a few trees, the sticky bands described above for monitoring may serve as sufficient controls. For more information on the biology and host preference of this beetle, see Johnson and Lyon (1988) and Hall (1986).

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Exotic Weeds I

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for kudzu, saltcedar, and Brazilian pepper. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

This module discusses the biology and management of three woody or semi- woody exotic weeds--kudzu, saltcedar, and Brazilian pepper--that are most abundant and damaging in the mid-Atlantic and southern United States. Due to their aggressive growth habits, these weeds outcompete and displace native plants. In addition they overgrow and damage structures, impede waterways, and may have direct toxic effects on animals. The management objectives for these three weeds differ according to the use of the affected land area, and range from local elimination of small or newly-established infestations to reductions of well-established populations to tolerable levels.

In some cases exotic vegetation is allowed to remain because it is historically accurate and contributes to the character of a cultural landscape. For example, some introduced species were brought to an area during a certain time period or by a particular group and thus provide important information about the history of a site. Although historically correct, these species can become an immense problem if they are not kept from spreading. Many historic sites have fallen into disrepair, allowing introduced plant species to spread into natural zones and force out native vegetation. Natural resource managers, cultural resource managers, and maintenance personnel must work together to establish priorities for the preservation of historic landscapes that consider protection of both the cultural and natural resource.

IDENTIFICATION AND BIOLOGY OF KUDZU, SALTCEDAR AND BRAZILIAN PEPPER

Kudzu

A native of Asia, *Puearia lobata* was introduced into the United States at the Philadelphia Centennial Exposition in 1876. Beginning in 1933, farmers in the South were encouraged to plant kudzu to reduce soil erosion. By 1953, it had become such a weed problem that it was removed from the USDA's list of permissible cover plants. In 1970, the USDA began listing kudzu as a common weed in the south. Today, kudzu is common in Alabama, Georgia, Mississippi, Tennessee, the Carolinas, Kentucky, Virginia, Maryland, and west to Texas and Oklahoma (Edwards 1982). The weed has also been reported in New York, Illinois, Iowa, Nebraska, and Washington (Shurtleff and Aoyagi 1977). It has also been observed at Biscayne and Everglades national parks in Florida.

P. lobata (Willd.) Ohwi is a legume of the subfamily Fabaceae. It is a trailing or climbing semi-woody perennial vine reaching 32'-100' in length. Young vines are covered with soft, fine hairs. First-year vines may reach 1/2" in diameter; old vines may reach a diameter of 4". As many as 30 vines may radiate from a single crown. Vines can grow up to 60' in a single growing season (and reportedly up to 1' per day). Vines may climb vertically as high as 50', completely covering trees, buildings, or other supporting objects. During the growing season, vines are densely covered by foliage. Leaves are alternate and compound, with three broad leaflets up to 4" across, each leaflet entire or deeply two or three lobed and with hairy margins. Foliage drops after the first fall frost. The roots of kudzu are fleshy; the taproot may reach over 6' in length, 7" in diameter, and may weigh up to 400 lbs.

Kudzu plants are perennial and do not usually flower until their third year. Flowers are purple, fragrant, about 1/2" long, produced in long racemes resembling pea flowers in shape. They are produced in August and September. Flowers are followed by flattened, 2" long hairy pods which may contain 3-10 hard seeds. Seeds are rarely produced in the United States (except on plants supported vertically on buildings, trees, or other supports [Shurtleff and Aoyagi, 1977]). In the United States, kudzu generally spreads by means of stolons (runners) and rhizomes. In addition, any vine contacting the soil will produce roots at nodes; these roots enlarge, forming new crowns. Vine cuttings and root divisions will also sprout. Vine nodes that come in contact with soil root to establish new plants; these roots produce new crowns, and the connection to the mother crown dies within one year after rooting. Kudzu is deciduous; its leaves drop after the first frost, and new leaves are produced each spring. See Shurtleff and Aoyagi (1977) or other weed atlases for drawings of kudzu.

Kudzu grows well under a wide range of environmental conditions, although best growth is achieved where winters are mild (40^{o}-60 ^{o}F), summer temperatures rise to about 80^{o}F, and rainfall is abundant (40" or more). Kudzu can grow in nearly any type of soil (e.g., acid soils, lime soils, lowlands with high water tables, and over heavy subsoil), and where winter soil temperatures remain above -25^{o}F (lethal temperature for roots). Forest edges or disturbed areas such as abandoned fields and roadsides are preferred habitats. Vines are intolerant of shade and grow toward light. Large roots store water, allowing plants to survive in fairly dry climates (to 20" of rain per year). Growth is most rapid in acid to neutral soils (pH 4.5-7.0).

Kudzu grows rapidly, choking out competing vegetation in sunny areas. Climbing vines may completely cover and shade out trees, and may cover and damage buildings, overhead wires, and other structures.

Saltcedar

This term includes *Tamarix* spp., especially *T. ramosissima* (Ledeb.), which is generally (but incorrectly) known as *T. pentandra* (Baum 1978). Saltcedar is a native of Eurasia and Africa, was introduced into the United States as an ornamental shrub in the early 1800s, and has now spread throughout the inter-mountain region of the western United States (Carman and

Brotherson 1982). Saltcedar is considered beneficial in that it provides good nesting habitat for wildlife (including doves) and is an excellent source of nectar for honeybees in Arizona and New Mexico (Deloach 1989).

Saltcedar is a deciduous shrub or small tree growing to 12'-15' in height. Slender, long gray-green branches are spreading or upright, often forming dense thickets. Scalelike leaves are gray-green, alternately arranged, narrow, pointed, about 1/16" long, and overlap one another on the stems. Active growth occurs from early or mid-spring to fall, when leaves drop. Leaves often become encrusted with salt secretions. Branches take on a brown-purple color as they age. Bark is reddish-brown and smooth on young branches, becoming ridged and furrowed on older limbs. Large numbers of pink to white flowers, about 1/16" across, appear in a dense mass on 1/2"- 2" spikes at branch tips from March to September. Flowers are pollinated by bees and other insects and produce greenish-yellow to pinkish-red capsules, 1/8"-1/5" long, which split into three to five parts on maturity. Seeds are 1/25" long, with a tuft of fine hairs at one end. The number of seeds per capsule is not constant. Seeds are dispersed by wind to new locations. Seedlings require extended periods of soil saturation for establishment. See Baum (1978) or Parker (1972) for drawings of saltcedar.

Saltcedar occurs in moist rangeland and pastures, bottomlands, banks, and drainage washes of natural or artificial waterbodies, and in other areas where seedlings can be exposed to extended periods of saturated soil conditions for establishment. Saltcedar can grow on soils with up to 15,000 ppm soluble salt. Established plants have long roots that can tap deep water tables. Furthermore, saltcedar has the highest known evapotranspiration rate of any desert phreatophyte (Carman and Brotherson 1982), which may result in water depletion from the underlying soil.

Among the serious direct impacts of this species are the displacement of native range plants by its aggressive growth, the possibly serious depletion of ground water due to its rapid evapotranspiration rate, increased deposition of sediments in tamarisk- infested streams, and the blockage of streams and artificial water channels by dense clumps of saltcedar growth, which can promote flooding during periods of heavy rain.

Brazilian Pepper

Brazilian pepper (*Schinus terebrinthifolius* [Raddi]) is a member of the Anacardiaceae, and is closely related to poison ivy. This weed was introduced from its native Brazil in 1898 by a USDA plant explorer (Morton 1978). It was considered an ornamental shrub and was distributed by the USDA Plant Introduction Station in Miami, FL, to local plant enthusiasts. Since then, Brazilian pepper has spread over thousands of acres of land in south and central Florida, the Florida Keys, the Hawaiian Islands, southern Arizona, and southern California.

Brazilian pepper is a broad-topped, rapidly-growing, evergreen tree reaching up to 40' tall, with a short trunk up to 40" thick. The trunk is usually hidden by a dense head of intertwining, contorted branches. Leaves are evergreen, pinnate, and have reddish midribs which may be winged. Each leaf bears 3-13 sessile, oblong or elliptical, finely toothed, glossy, resinous, aromatic 1-2" leaflets. These are dark green on the top and lighter on the underside. Five-petaled, white, 1/8" flowers are borne in 6" sprays originating in leaf axils along the upper 32"-43" of each stem. Male and female flowers are borne on separate trees. Flowering peaks in October in Florida. Blooms are followed by masses of round single-seeded drupes, which change from green to bright red at maturity. The appearance of the fruit is responsible for the common names "Florida holly" and "Christmas berry." Seeds may be dispersed by birds or small mammals or may germinate near the parent plant, producing dense spreading colonies. See Olmsted and Yates (1984) for photographs of Brazilian pepper.

This tree quickly colonizes disturbed areas. Seedlings can tolerate low light levels, growing slowly until the overstory canopy is opened up. Dead trees should be allowed to remain in the canopy to provide as much shade as possible. Seedling survival is low on inundated ground. Trees can withstand extended drought, and up to six months of inundation. Large trees can withstand fires and high winds without suffering significant damage (Olmsted and Yates 1984). Apparently, Brazilian pepper can tolerate Mediterranean, tropical, and desert climates.

Direct negative environmental impacts include the displacement of native plants, not only because of this species' aggressive, rapid growth, but also because of allelopathic effects (toxic or inhibitory activity) of chemicals in vegetative plant parts and fruits. Brazilian pepper is closely related to poison ivy and can produce effects similar to that plant on humans and animals (Lloyd et al. 1977; Morton 1978; Olmsted and Yates 1984). Massive bird kills in Florida may have been caused by excessive feeding on Brazilian pepper berries.

MONITORING AND THRESHOLDS

Kudzu

Regular monitoring of both developed and natural areas is required to determine the presence and extent of kudzu incursions. Since this species is a rapid grower and an aggressive competitor, these inspections should be conducted frequently (at least monthly) during the growing season.

In addition to inspecting areas for actively growing kudzu, monitors should also inspect disturbed areas, which can be rapidly colonized by the weed. All records of sightings of kudzu and of disturbed sites should be recorded, maintained, and updated at each inspection.

Since this weed is an adaptable, aggressive competitor that can rapidly overgrow native vegetation, the presence of any kudzu should trigger control activities. There is no acceptable population level (L.K. Thomas, Jr., personal communication).

Saltcedar

Inspection of both developed and natural areas is necessary to determine the presence and extent of saltcedar incursions. One inspection should be made early in the growing season (before or at flowering), to identify mature plants and initiate control before seed can be set and distributed. Additional inspections should be made later in the growing season to identify seedlings developing from seed set in the current year. All records should be maintained and updated at each inspection.

The presence of any saltcedar should trigger control activities, although it should be recognized that where stands are extensive, elimination is probably infeasible (P. Sanchez, personal communication).

Brazilian Pepper

Inspection of all likely habitats is required to determine the presence and extent of Brazilian pepper incursions. At least one inspection per year should be made for the presence of established plants. Frequent inspections (at least monthly) should be made for the presence of disturbances in the normal plant cover (e.g., due to storms, alterations of water levels, fires, and human activities), since such sites can be rapidly colonized by this weed. All records should be maintained and updated at each inspection.

The presence of any plants should trigger control activities, since this species is capable of displacing native vegetation.

NON-CHEMICAL CONTROLS

Kudzu

Cutting. Vines (including runners) are chopped just above ground level and the pieces destroyed by burning or feeding to livestock. Early in the season, cutting is repeated at two-week intervals to weaken the crown and prevent resumption of photosynthesis. Later in the season, when the stored energy in the taproot has been reduced, the interval between cuttings can be extended (L.K. Thomas, Jr., personal communication). Cutting does not affect roots or crowns, which will regrow unless their supply of stored energy is depleted.

Flaming. A kerosene torch is played over the foliage, wilting the leaves and defoliating the plant. Flaming should be done according to the same schedule as cutting. Where all foliage can be reached, this method may be more effective than cutting. Like cutting, flaming does not affect the roots or crowns.

Burning. Destroys above-ground growth. Since kudzu vines usually will not burn during growth (because of their high water content), vines may be flamed (see above) two or three days prior to burning. This causes the leaves to wilt and dry, providing fuel for the burning process.

Grubbing. This consists of mechanical removal and destruction of the entire plant, including the taproot. If all root tissue is removed, no regrowth can occur, so repetition should not be necessary. However, this procedure can be destructive to the treated area. Removal of crowns only is more effective than cutting, but must be repeated, since remaining roots will re-sprout. Crown removal is most effective at flowering (when the plants are weakest) or in the fall. However, the crowns are difficult to find except in the spring, when the operation will be less effective.

Grazing. Kudzu is a favored food of goats and cows, which can provide useful levels of control.

Where these can be accommodated in the park management plan, this technique can be effective.

Saltcedar

Cutting. This process involves removal of all growth at ground level, but regrowth is not prevented.

Burning. This removes above-ground growth, but allows remaining roots and crowns to re-sprout.

Grubbing. Grubbing with a grubber blade, which is smaller than a root plow, is used to remove smaller stands. This is less destructive than root plowing.

Root pulling. Removal of the main portion of the root system and crown is labor and time intensive. Regrowth from incompletely-removed roots may occur.

Chaining. A chain, 360'-400' long, and weighing 40-50 lb/ft., can be doubled and pulled between two crawler tractors. Chaining may uproot whole plants or may shear trunks at ground level. Drawbacks of chaining include the failure to remove all below-ground tissue, allowing regrowth as well as the destructiveness of the procedure itself.

Root plowing. This process shears vegetation below the ground surface. The root plow kills medium to large shrubs by shearing below the crown, largely (but not completely) preventing regrowth. This technique is destructive to the environment but is widely used in areas where saltcedar coverage is nearly 100% (Gangstad 1982).

Drag lining. Drag lines are used to shear vegetation growing in water bodies or channel banks. It is not suitable for large vegetation.

Bulldozing. This shears plants at ground level, or uproots entire plants. Regrowth from sheared trunks can occur. This, also, is a destructive technique.

Inundation. Flooding can be used to control saltcedar growing on lake shores if root crowns can be flooded for at least three months (DeLoach 1989).

Brazilian Pepper

Hand removal. Seedlings or small saplings can be pulled from the soil. Pulled plants must be removed from their growing site and bagged or dried to prevent re- sprouting.

Burning. Olmsted and Yates (1984) report that prescribed burning has kept a slash pine forest in Florida free of Brazilian pepper seedlings.

Bulldozing. This technique has been used in the Everglades National Park (Olmsted and Yates 1984).

BIOLOGICAL CONTROL

Kudzu

In the United States, kudzu vines may be attacked by a root knot nematode (*Meloidogyne* sp.), a "blackleg" fungus disease, a viral mosaic disease, and a rust fungus (Shurtleff and Aoyagi 1977). These pests produce only minor injury and are not known to kill kudzu plants.

Saltcedar

Watts et al. (1977) found only a few native insects that fed on saltcedar in New Mexico. These did little harm to the plants except under exceptional circumstances. Aphids, grasshoppers, beetles, and spider mites were among the organisms found. Watts et al. also reported two introduced insects, the leafhopper *Opsius stactogalus* and the scale insect *Chionaspis etrusca*, were found regularly on saltcedar. The leafhopper sometimes caused substantial damage. Baum (1978) compiled a list of insects and fungi that attack various species of *Tamarix* in Europe, Africa, and Asia, but found no records of enemies of *T. ramosissima*. Deloach (1989) recently reviewed the prospects for biological control of saltcedar and suggested that through the importation of several biological control agents from Asia and other areas, control of this weed could approach 80%.

Brazilian Pepper

Goats can graze on foliage of this species without suffering ill effects (Morton 1978). A witches' broom disease fungus, *Sphaeropsis tumefaciens* Hedges, attacks Brazilian pepper, but is also a pest of *Ilex opaca*, *Citrus* spp., and numerous ornamentals. The red-banded thrips (*Selenothrips rubrocinctus* Giard.) occasionally kills plants, but is also a pest of mango and cashew plantings. Recently, Bennett et al. (1989) reviewed the status of biological control activities directed against Brazilian pepper. Three species of insects were recently imported to Hawaii from South America for the control of Brazilian pepper. A bruchid beetle, *Bruchus atronotatus*, and a tortricid moth, *Episimus utilis*, established on the weed but caused no significant population reductions. A gelechiid moth, *Crasimorpha infuscata*, apparently failed to establish (Bennett et al. 1898).

CHEMICAL CONTROLS

Several herbicides have been used to manage the weeds described in this module. These will not be described in detail, since new herbicides are constantly being developed. Contact your regional Integrated Pest Management coordinator for information on the most appropriate materials to use in your situation.

Kudzu

Several herbicides have been successfully used in the National Park Service to manage kudzu.

These projects are described in Bratton (1981), Rosen (1982), and Gangstad (1989). Cut stump treatments have been effective at Great Smoky Mountains National Park. These work best on small infestations or after foliar treatments the previous season.

Saltcedar

A variety of herbicides have been used to manage saltcedar. These are generally applied as cut-stump treatments, although foliar, stem-sprout, root-sprout, injection, frill, and broadcast applications are used as well. When cut-stump treatments are used, the herbicide should be in a non-evaporative base so that the stump does not dry out before the chemical has entered. Deloach (1989) has reported on successful use of this technique. Gangstad (1989) has also described several methods for chemical control of saltcedar.

Brazilian Pepper

Non-woody seedlings can be treated with foliar applications. Small woody saplings and established trees can be treated with a spray to every major stem (complete coverage to runoff, at 12"-15" above ground level). Treated sites should be monitored and surviving trees retreated at six week intervals following treatment, until regrowth no longer occurs. Gangstad (1989) described a technique for management of Brazilian pepper in rangeland and permanent pastures.

SUMMARY

Kudzu

Regular cutting (or flaming, where applicable) may be sufficient to control most kudzu populations. Grubbing may control small infestations, if it will not result in too much destruction of other vegetation. Where it can be accommodated, grazing by goats may preclude the need for additional measures. For large overgrown areas, application of a recommended pesticide may be necessary.

Saltcedar

Individual plants can be grubbed from the soil. Cutting followed immediately by application of herbicide to stump ends is the most effective means of controlling small stands of mature shrubs.

Brazilian Pepper

Small trees or individual seedlings can probably be mechanically pulled by workers wearing protective clothing. Prescribed burns may prevent establishment of seedlings in appropriate circumstances. Cutting and bulldozing may be useful against large trees and stands. Seedling stands and established trees may be treated with registered herbicides.

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PERSONAL COMMUNICATIONS

L.K. Thomas, Jr.
Research Biologist
Center for Urban Ecology
National Capital Region
National Park Service
Washington, D.C.

P. Sanchez
Research Manager
Haleakala National Park
National Park Service
Hawaii

Exotic Weeds II

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for tree of heaven, Japanese honeysuckle, mimosa tree, siris tree, giant sensitive plant, and sensitive plant. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

This module discusses the biology and management of six woody weeds that are either trees, shrubs, or vines and are distributed throughout many regions of the United States. They are pests because they are invasive and can displace native vegetation and because they create floristic inaccuracies in historical landscapes due to their foreign origin. One is an alternate host to an important nematode pest of agronomic crops.

In some cases exotic vegetation is allowed to remain because it is historically accurate and contributes to the character of a cultural landscape. For example, some introduced species were brought to an area during a certain time period or by a particular group and thus provide important information about the history of a site. Although historically correct, these species become an immense problem if they are not kept from spreading. Many historic sites have fallen into disrepair, allowing introduced plant species to spread into natural zones and force out native vegetation. Natural resource managers, cultural resource managers, and maintenance personnel must work together to establish priorities for the preservation of historic landscapes that consider protection of both the cultural and natural resource.

IDENTIFICATION AND BIOLOGY OF TREE OF HEAVEN, JAPANESE HONEYSUCKLE, MIMOSA TREE, SIRIS TREE, GIANT SENSITIVE PLANT, AND SENSITIVE PLANT

Tree of Heaven

Tree of Heaven (*Ailanthus altissima* [Miller])is in the family Simaroubaceae. It was introduced into the United States from Asia (China) as a host tree for the Cynthia moth, *Samia cynthia* (Drury), which was introduced for silk production. It was brought to the eastern United States as nursery stock because of its ability to grow quickly under adverse conditions. Chinese miners also brought the seeds with them to California because of their medicinal and cultural importance. Distribution in the United States is from Massachusetts to Iowa and Kansas and south to southern Texas and Florida. Tree of heaven has established to a lesser extent in the western United States from southern Rockies to the Pacific Coast states. It is a tall (to 60'), deciduous, polygamous tree and often colonizes by root sprouts. Stump sprouts can grow 6'-12' in length in a single summer. Flowers are present in late May through early June in 12" long terminal panicles. A large cluster of pink fruits develops from July to October. The flowers and vegetative parts, if bruised, are ill scented, almost nauseating, on hot days.

Tree of Heaven is intolerant of deep shade and occurs most commonly along fence rows, roadsides, and waste areas. It is tolerant of urban conditions, including compacted, poor soils, and polluted air, and is common in dusty, smoggy areas such as inner cities where most other trees fail. It is often used as an ornamental in urban areas. It spreads rapidly in disturbed areas and can quickly take over forest openings created by gypsy moth damage or fire. It can pose a serious threat to natural areas. It has been found growing up to two air miles from the nearest seed source.

Japanese Honeysuckle

Japanese honeysuckle (*Lonicera japonica* [Thunberg]) is a high climbing or trailing vine in the family Caprifoliaceae. It was introduced into the United States from Asia. Distribution in the United States is from the central Atlantic states to Missouri and Kansas, south to Florida and Texas. Japanese honeysuckle stems are glabrous to densely pubescent. Leaves are 3/4"-2 1/2" long, evergreen, oval in shape, with a rounded base. In spring the leaves of new shoots are often lobed. The flowers are very fragrant and occur in pairs. They are white or pink when they first appear and fade to yellow with age.

This vine, originally planted as an ornamental and to stabilize road banks, has invaded woodlands, fence rows, and fields, outcompeting and killing native wild flowers, shrubs, and tree seedlings. It is common to abundant at low altitudes, but can spread into uplands. It grows best in full and partial sun but t olerates partial shade. This species is considered a major pest due to its ability to outcompete and shade out native vegetation.

Mimosa Tree, Powder-puff Tree, Silktree, Mimosa

Mimosa tree (*Albizia julibrissin* [Durazzini]) as introduced into the United States as an ornamental from Asia and Africa. Distribution in the United States is from the mid-Atlantic states south, and as far west as Indiana. It is a flat-topped, thornless, deciduous tree which reaches 35' in height.

The mimosa tree was introduced as an ornamental but escaped into fields and waste areas. It does not establish in forests, but commonly occurs on forest borders. It can also invade riparian areas and spread downstream. It is o ften injured by severe winters. Its major negative impact is its improper occurrence in historically accurate landscapes.

Siris Tree, Woman's Tongue

Albizia lebbeck (L.) Benth. (family Leguminosae) was introduced from Egypt into southern Florida about 1900 where it escaped from cultivation. It is probably a native of tropical Asia, but is widely planted throughout the tropics as a shade tree and ornamental. Its range extends to Bermuda and the West Indies, Central America, and south to Brazil. It is a medium-sized deciduous tree 20'-40' high, to 1/2' in diameter or larger, with a spreading crown of thin foliage. The bark is gray, and smooth but becomes rough as the tree ages. The fruits are flat pods, which are broad, straw- colored, 4" to 8" or more in length, and 1" to 1/2" wide. They usually occur in large numbers. Pods remain on the tree for some time after the seeds and leaves have fallen. The sound of the empty pods rattling in the wind gives the tree its common name of woman's tongue.

This species propagates readily from seed and has established in pastures and on hillsides in dry coastal regions. This species is highly tolerant of salt spray but is intolerant of cold temperatures.

Giant Sensitive Plant

The giant sensitive plant (*Mimosa invisa* [Mart.]) is a native of Brazil. This species is a weed in many tropical and subtropical countries and is found in the United States in Hawaii. An erect, climbing shrub, the giant sensitive plant is biennial or perennial depending on the climate, and often forms dense thickets (Holm et al., 1977). This species possesses a strong root system that often becomes woody at the base. Stems can be up to 6' tall, and are conspicuously angular, with many randomly scattered recurved spines or thorns 1/4"-1/2" in length. The seeds are flat, about 1/4" long, and are adapted for dispersal by animals. Seedlings only a few weeks old may produce viable seeds. Some of these may germinate immediately while others may remain in the soil for several years before germination. See Holm et al. (1977) for illustrations.

Often found in moist waste places, plantations, pastures, and cultivated areas, this plant has become a serious weed in sugar cane plantations. It covers other vegetation with the spiny stems, forming spreading tangled masses or impenetrable thickets up to 6' high.

This species has been designated a noxious weed by the United States Department of Agriculture (Westbrooks 1981). It overruns and outcompetes native vegetation in large areas. It further affects native species by being unpalatable to most grazers and by trapping animals caught in thickets; animals or people may die or become seriously injured if they become ensnared in these thickets. Animals will not browse or step on stems due to the recurved thorns (Holm et al. 1977).

Sensitive plant

The sensitive plant (*Mimosa pudica* [L]) is a multistemmed, perennial shrub in the legume family. Originally from tropical America, the sensitive plant is widely introduced and is now found throughout the New World tropics. It is considered a troublesome weed in the Caribbean region and South America. In the United States its distribution is limited to Hawaii and Puerto Rico. Its bristled seeds are dispersed by attachment to animals or people. Seeds may remain in the soil for several years before germination; seeds stored under laboratory conditions have shown 2% germination rates after 19 years (Holm et al. 1977). See Holm et al. (1977), Radford et al. (1964), Fernald (1950), Little (1953), Little and Wadsworth (1964), and Gleason (1963) for more detailed descriptions of this species.

Sensitive plant is found in cultivated areas, lawns, and waste places. It grows on a wide variety of soils and has a high tolerance for shade. It is often grown as an annual ornamental for its showy flowers and as a cover crop in some tropic countries for its nitrogen-fixing abilities. It occurs as a common weed in many cultivated crops and in pastures, where its high populations and thorny stems make grazing difficult. Sensitive plant is an alternate host to several species of parasitic flowering plants and of the root-knot nematode *Meloidogyne* sp., which is a serious pest of many crop plants (Holm et al. 1977).

MONITORING AND THRESHOLDS

Monitoring techniques for the introduced weed species described above consist of periodic visual inspections. All observations and treatments should be recorded.

Care should be taken to monitor small, slowly expanding populations which have not reached pest status. A slight change in environmental conditions, such as drought or fire, could enable populations to grow rapidly (Anonymous 1983). Control efforts should be undertaken whenever any of these species is observed, as they can all spread rapidly and overtake desired vegetation.

NON-CHEMICAL CONTROLS

Tree of Heaven

Cutting. This process involves removal of all above-ground growth. Regeneration of stump sprouts and from underground parts is not prevented.

Japanese Honeysuckle

Cutting. Vines may be chopped just above ground level. Cutting is repeated every two weeks to deplete nutrient reserves in the roots and prevent resumption of photosynthesis. Cutting does not affect roots, which will continue to grow until their energy and nutrient supplies are depleted.

Flaming. By placing a kerosene torch over leaves on the same schedule as cutting, foliage is wilted and nutrient supplies in the roots are depleted. As with cutting, flaming will not affect roots.

Burning. Although few quantitative studies occur in the literature, Barden and Matthews (1980) recommend controlled burning. Two annual burns in an experimental plot reduced honeysuckle crown volume by 80%. Ground cover was reduced by 35%. Fires killed most above-ground vines, but ground cover was maintained by re- sprouting roots. Burning may be combined with previous flaming, which wilts and dries leaves, providing fuel for the burn.

Grubbing. Consists of mechanical removal and destruction of the entire plant, including the root. If all root tissue is removed, no regrowth can occur, and repetition is not necessary. Grubbing is labor intensive and may be locally destructive. Grubbing is most effective from fruiting to winter and early spring when plant reserves are lowest.

Grazing. Controlled grazing by goats may serve to reduce honeysuckle crown and ground cover densities, but as with controlled burning, re-sprouting roots will regenerate unless nutrient reserves are depleted by continuous grazing pressure.

Regardless of the control method used, care must be taken to prevent re- invasion from nearby areas, or by seeds transported by birds or other wildlife. Planting the area with fast-growing native vegetation or grasses may prevent recolonization.

Mimosa Tree

Cutting. See Tree of Heaven.

Siris Tree

Cutting. See Tree of Heaven.

Giant Sensitive Plant

Controlled burning. See Japanese honeysuckle.

Sensitive Plant

Controlled Burning. See Japanese honeysuckle.

BIOLOGICAL CONTROL

Tree of Heaven

The Cynthia moth, *Samia cynthia*, feeds on this species, but it is rare outside of urban habitats (Pyle 1983).

Japanese Honeysuckle

No natural enemies are reported for this species.

Mimosa Tree

Attacked by mimosa wilt, *Fusarium oxyapoeum perniciosum*, a fungus. It is also fed upon by the mimosa webworm, *Homadaula anisocentra* Meyrick, and the root-knot nematode *Meliodogyne incognita*.

Siris Tree

No natural enemies are reported for this species.

Giant Sensitive Plant

No natural enemies are reported for this species.

Sensitive Plant

No natural enemies are reported for this species.

CHEMICAL CONTROL

Tree of Heaven

Current treatment consists of felling and stump treatment with herbicide. Chemical treatment kills remaining tissue and prevents regrowth of stump sprouts. Trees may be frilled and treated with felling, treated by injection, or treated by hack and squirt. The latter technique involves cutting into the cambium and applying a herbicide into the wound.

Honeysuckle

Mclemore (1981) reports that an acceptable level of control (70%) was reached during a two-year experimental program which used 2 lb/acre of glyphosate in the first year and 6 lb/acre in the second year. Japanese honeysuckle is an evergreen, so it can be treated in the dormant season with less damage to non-target species.

Mimosa Tree

Use the same treatments described in the Tree of Heaven and Honeysuckle sections.

Siris Tree

Everglades National Park has successfully used basal bark sprays of herbicide for management of this plant. It is applied to runoff to the complete circumference of the trunk 12" to 15" above the ground (Anonymous 1983). A carrier and dye are added to the herbicide to ensure good penetration and complete coverage by the herbicide.

Giant Sensitive Plant

See Sensitive Plant section.

Sensitive Plant

Patro and Tosh (1974) recommend postemergence application of 2,4-D as the best chemical control measure for this species.

Consult with your regional integrated pest management coordinator to determine which, if any, pesticide is best suited to your integrated pest management program.

SUMMARY

Tree of Heaven

Felling individual problem trees and treating the cut stumps with approved herbicides to prevent regrowth may be sufficient for control in most situations. Depending on the growth form of the plant, basal-bark treatments or foliar treatments with herbicide may be necessary as well.

Japanese Honeysuckle

Regular cutting (or flaming, where applicable) followed by spot treatments of herbicides or regular controlled burns combined with spot treatments or grazing pressure may control honeysuckle in most situations. Grubbing or other mechanical methods should be sufficient for small infestations.

Mimosa Tree

See Tree of Heaven section.

Siris Tree

See Tree of Heaven section.

Giant Sensitive Plant

Flaming, burning, and postemergence applications of herbicide followed by spot treatments with approved herbicides may be sufficient for control in most situations. Broadcast treatments with approved herbicides may be required to treat large infestations.

Sensitive Plant

See Giant Sensitive Plant section.

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Fire Ants

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for fire ants. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Fire ants are so called because their venom, injected by a stinger like a wasp's, creates a burning sensation. They are also active and aggressive, swarming over anyone or anything that disturbs their nest, be it wild animals, domestic animals, pets or people. An encounter with a fire ant nest can leave a lasting memory of burning pain, followed by tiny, itching pustules.

Because of this, and occasional stories of animals or people killed by multiple stings, people fear fire ants. In some areas infested with certain species of fire ants, playgrounds, parks, and picnic areas lie abandoned, unused because of the presence of fire ants. In campsites of state and national parks in fire ant infested areas, it is often difficult to put up or take down a tent without being stung by angry fire ants.

Fire ants are pests in other ways besides their stinging. They damage crops such as soybeans, eggplant, corn, okra, strawberries, and potatoes by feeding directly on the plants or by protecting other insects that damage the crops. They chew the bark and growing tips of citrus trees and feed on the fruit. Fire ant mounds interfere with farming and mowing operations and turn recreational fields into disfigured moonscapes. Fire ants have caused sections of roads to collapse by removing soil from under the asphalt.

Increasingly, fire ants have been found nesting in wall voids, around plumbing, and under carpeting in structures. The ants have also been found invading outdoor electrical equipment, apparently attracted to the electrical fields. Infested sites include household electric meters, traffic signal control boxes, and even airport runway lights.

Fire ants are voracious predators and sometimes feed on pests such as boll weevils, sugarcane borer, ticks, and cockroaches. The imported fire ant is thought to have dramatically reduced the range of the lone star tick, a serious livestock pest.

BIOLOGY AND IDENTIFICATION OF FIRE ANTS

Pest Species of Fire Ants

There are many species of fire ants in the United States, but the most serious pests for National Park Service personnel are four in the genus *Solenopsis*: the red imported fire ant, the black imported fire ant, the southern fire ant, and the fire ant. Distinguishing between imported and native species of fire ant is difficult, even for experts. Identification usually requires 40 or more randomly collected worker ants for study. The following sections describe the four fire ants of major concern.

Red Imported Fire Ant

Introduced from South America, this species becomes the number one fire ant pest wherever it occurs. The red imported fire ant (*Solenopsis invicta*) is associated with disturbed habitats, mostly created by humans, and is abundant in old fields, pastures, lawns, roadsides and many other open sunny areas. It often inhabits fields used for agricultural purposes where its large above-ground mounds create problems in planting and harvesting crops. In areas where grass is periodically cut, mounds are flush with the ground and are hard to see. This species is rarely found in mature forests and other areas with heavy shade, unless part of the area has been disturbed by fire or storms.

The red imported fire ant builds mounds that are, on average, 10"-24" in diameter and 18" high. But larger mounds are not uncommon. They also may extend 6' underground. The primary function of mounds, beyond that of the simple ground nests of other ants, is microclimate regulation--controlling the temperature and humidity. The ants can maintain a temperature inside the mound much higher than that outside, allowing them to continue colony growth during cool weather.

The mounds are symmetrical piles of excavated soil, rich in organic materials, laced with interconnected galleries and chambers. The soil below ground also contains galleries and chambers. During foraging periods only a small percentage of ants may be inside the mound; the rest are out gathering food and exploring.

A newly established nest rapidly produces young, and winged reproductives are produced for most of the year (8-10 months), much longer than native species. Red imported fire ants quickly spread through a suitable habitat, and the species is now found throughout most of the southeastern United States and west into Texas.

Black Imported Fire Ant

The black imported fire ant, *Solenopsis richteri*, is very similar to the red imported fire ant. It is currently limited to a small area of northern Mississippi and Alabama. It may be displaced from established habitats by the red fire ant.

Scientists have long thought that the black and red fire ants were two distinct species. Recently it has been discovered that hybrids of these ants produce viable offspring, and some scientists now wonder whether they are simply two races of the same species, varying in color and perhaps behavior.

Southern Fire Ant

The southern fire ant, *Solenopsis xyloni*, is a native species that occurs from North Carolina south to northern Florida, along the Gulf Coast and west to California. Colonies may be observed as mounds or more commonly may be constructed under the cover of stones, boards, and other objects or at the base of plants. These ants also nest in wood or the masonry of houses, especially around heat sources such as fireplaces. Nests often consist of loose soil with many craters scattered over 2 to 4 square feet. In dry areas nests may be along streams, arroyos, and other shaded locations where soil moisture is high. Southern fire ants usually swarm in late spring or summer.

Fire Ant

The fire ant, *Solenopsis geminata*, is a native species sometimes called the tropical fire ant. This ant ranges from South Carolina to Florida and west to Texas. Very similar to the southern fire ant, it usually nests in mounds constructed around clumps of vegetation, but may also nest under objects or in rotting wood.

The Ant Colony and Life Cycle
The life cycles of the four fire ant species discussed above are very similar.

Development of the individual: Like all ants, an individual fire ant begins life as an egg, which hatches into a legless, grub-like larva. The larva is very soft and whitish in color. It is also helpless and depends totally on worker ants for food and care. The larva is specialized for feeding and growing, and almost all growth occurs during this period. As in all insects, growth is accomplished by periodic molting, or shedding of the cuticle (skin). Having reached its final size, the larva becomes a pupa in which various adult structures, such as legs, and in some cases wings, become apparent for the first time. The pupal stage is the transitional stage between the larva and the adult that emerges during the final molt. In insects in general, the adult stage is specialized for reproduction and dispersal; with ants, some adult individuals are capable of reproduction (queens and kings) and the remainder are sterile workers.

The colony: The social unit of fire ants is the colony. Colonies, like individuals, pass through a characteristic life cycle.

Fire ants are very typical of ants in general. In addition to workers and a queen, mature colonies contain males and females capable of flight and reproduction. These individuals are generally called "reproductives." On a warm day, usually one or two days following a rain, the workers open holes in the nest through which the reproductives exit for a mating flight. Mating takes place 300' to 800' in the air. Mated females descend to the ground, break off their wings, and search for a place to dig the founding nest, a vertical tunnel 2" to 5" deep. They seal themselves off in this founding nest to lay eggs and to rear their first brood of workers. During this period they do not feed, instead utilizing reserves stored in their bodies. The first worker brood takes about a month to develop; these are the smallest individuals in the entire colony cycle. They open the nest, begin to forage for food, rear more workers, and care for the queens. Hereafter, the queen or queens essentially become egg-laying machines, each able to lay up to 1,500 eggs per day.

Multiple queen colonies are fairly common. A single colony may have 10 to 100 or more queens, each reproducing. Multiple queen colonies can mean up to 10 times more mounds per acre. The queens generally mate several times and may live for several years. Workers are less long-lived and usually will not survive an entire season.

The colony grows rapidly by the production of workers that gradually enlarge the original vertical tunnel into multiple passages and chambers. Colony maturity is attained when reproductives are once again produced. The reproductives leave to mate and form new colonies. A mature colony of red imported fire ants can produce as many as 4,500 reproductives during the year in 6-10 mating flights between spring and fall. Nearly 100,000 queens may be produced per acre in heavily infested land, but mortality rates, mostly from predators, can reach 99%.

Colony size: Colonies of red and black imported fire ants become territorial as they grow; they defend an area against all other fire ants. Therefore, fire ant colony populations often reach an upper limit depending on the territory size of mature colonies. A typical figure for pasture land seems to be about 20-50 mounds per acre in single queen nests and up to 250 mounds or more in multiple queen nests. Mature colonies of imported red fire ants consist of an average 80,000 workers, but colonies of up to 240,000 and more have been reported.

Feeding Habits

The oldest and most expendable 20% or so of the colony's workers leave the nest to search for food. They explore 50-100 feet from the nest with an efficient looping pattern. Although the worker ants can chew and cut with their mandibles, they can only swallow liquids. When they encounter liquid food in the field, they swallow it and carry it back to the nest. Solid food is cut to reasonable size and carried back to the nest.

Like other ants, fire ant workers share their food with their nest mates by regurgitating it so that it can be licked or sucked by other ants. In this way, most ants in the nest get fed equally. This food sharing is also why baits can be an effective control tactic against fire ants.

Fire Ant Stings

In infested areas, fire ant stings occur more frequently than bee, wasp, hornet, and yellowjacket stings. Stepping on a mound is almost unavoidable when walking in heavily infested areas. Furthermore, many mounds are not easily seen, with many lateral tunnels extending several feet away from the mound just beneath the soil surface. Ants defend these tunnels as part of their mound.

A person who stands on a mound or one of its tunnels, or who leans against a fencepost included in the defended area, can have hundreds of ants rush out to attack. Typically, the ants can be swarming on a person for 10 or more seconds before they grab the skin with their mandibles, double over their abdomens, and inject their stingers.

Although a single fire ant sting hurts less than a bee or wasp sting, the effect of multiple stings is impressive. Multiple stings are common, not only because hundreds of ants may have attacked, but because individual ants can administer several stings. Each sting usually results in the formation of a pustule within 6 to 24 hours. The majority of stings are uncomplicated, but secondary infections may occur if the pustule is broken, and scars may last for several months. Severe infections requiring skin grafting or amputation have been known to occur.

Some people experience a generalized allergic reaction to a fire ant sting. The reaction can include hives, swelling, nausea, vomiting, and shock. People exhibiting these symptoms after being stung by fire ants should get medical attention immediately. Death can occur in hypersensitive people. Individuals who are allergic to fire ant toxins may require desensitization therapy.

Fear of Fire Ants
An important indirect effect of the presence of fire ants is fear of being stung. Fear and anxiety about fire ants may limit the use of sites where fire ants are present by park visitors and personnel alike. In some parks, playgrounds, athletic fields, and campsites are not used because of fear of fire ants in the area.

MONITORING AND THRESHOLDS FOR FIRE ANTS

Monitoring

The first step is to identify the species of fire ants in the area (see Pest Species of Fire Ants above). Population monitoring for fire ant control generally consists of determining the number of active mounds in a particular unit area. Any mound where at least three ants are observed after mound disturbance should be considered active. Heavily infested fields can contain over 100 active mounds per acre.

Another method of estimating ant populations for comparison studies is by collecting ants attracted at baits in a test area. A small piece of hamburger and a small piece of agar containing 40% honey are each placed on a small piece of aluminum foil or in a small plastic cup. The two baits are placed on the ground at each bait station, 1'-3' apart, at each bait station. Bait stations are placed about 10 yards apart. The number of ants attracted to the baits per unit time is determined.

Threshold/Action Population Levels

The threshold population levels for fire ants will vary according to the species and the sites. In certain camping and recreational areas, for example, very few active mounds per acre would likely be tolerated, particularly of the imported species. In contrast, a few active mounds per acre probably would be acceptable in other types of sites; little-used hiking areas, for example. Every effort should be made to correlate fire ant populations observed through the use of monitoring techniques with complaints received from park visitors and personnel. In this way, a complaint threshold level can be established for each park site.

In areas where fire ants are not causing any problems, the best solution may be to do nothing. Some sites will only support a limited number of fire ants. These may be in the form of a few large colonies or many small ones. Established mounds defend territories, preventing the establishment of new colonies. Maintaining several large, and perhaps well-marked, colonies may be a sound way to stabilize fire ant populations in an area, as long as there is a low risk of people or pets stumbling into the nest.

Some researchers believe it may be best to selectively control fire ant colonies- -allowing native species to flourish as a way to prevent the introduction of the imported species, or leaving single queen imports alone to prevent the area from invasion by a multiple-queen "supercolony."

Mounds built by fire ants in fields often interfere with mowing and farming operations. Not only is equipment damaged by dried and hardened fire ant mounds, but operators may refuse to enter fields infested by ants. The number of mounds per acre that can be tolerated as regards equipment damage must be determined on a case- by-case basis.

NON-CHEMICAL CONTROL OF FIRE ANTS

Fire ants, particularly red and black imported fire ants, pose a serious dilemma in parks. On the one hand, there can be no doubt that the fire ant is a major pest, stinging visitors and park workers, disfiguring the landscape, even attacking native animals. In one private preserve, imported fire ants were killing hatchlings of the brown pelican, a threatened species. On the other hand, aggressive insecticide treatment of critical habitat can have negative impact on a sensitive environment.

Fire ant management consists of a series of questions and decisions: What species are in the area? How extensive is the infestation? How high is the risk that visitors or park personnel will be stung? How much damage are the ants doing? Is control action justified? What are the best strategies of control? Answering these questions requires inspection and monitoring to determine the nature and extent of the problem.

Water

Boiling water has been added to individual mounds with varying degrees of success reported. Approximately 3 gallons of hot water poured into each mound will eliminate about 60% of the mounds treated. Surviving mounds will need to be treated again. Water has also been applied as steam, using a steam generator, usually on a cool day. Both techniques are cumbersome in the field, especially where large numbers of mounds are involved.

Area-wide flooding or prescribed burning of fire ant infested areas has proved ineffective, and may promote the establishment of new colonies.

Mechanical Disturbance

Mounds can be dug up and moved or destroyed, but not without some risk that the fire ants will successfully attack the digger. Dragging, or knocking down, mounds may provide a limited level of control, but only if mounds are dragged just before the first hard freeze. Mounds are destroyed by pulling a steel I-beam drag, weighing about a ton, behind a tractor across the ant-infested area. Destroying mounds during the warm season will not reduce the number of active mounds;

ants quickly rebuild their nests.

A number of mechanical mound pulverizers, ant electrocuters, even nest exploders, have been developed for fire ant control, but so far the effectiveness and practicality of these devices is open to question.

Biological Control

A number of biological enemies of the fire ants have been evaluated as biocontrol agents, including nematodes, bacteria, fungi, viruses, and microsporidia. Some show promise, but biological control is not yet a proven effective control tactic for fire ants.

So far, the most effective of these is a nematode, *Neoaplectana carpocapsae*. In trials, one application has inactivated about 80% of treated mounds in 90 days. The straw itch mite, *Pyemotes tritici*, has also been shown to inactivate fire ant mounds. Three to ten applications at about two week intervals gave 70% control. Practical use of this mite for fire ant control must await the development of more efficient methods of mass production and increased effectiveness. Another problem is that this mite is a pest of people and animals; it bites and causes a dermatitis.

Ant-Proofing

Fire ants, like other ants, may be nesting near buildings and can enter and move through a structure through innumerable tiny cracks and openings. Caulking and otherwise sealing cracks and crevices being used by fire ants can often have great effect in suppressing the population inside. Many effective, easy-to-use silicon sealers and expandable caulk products have been recently developed, including some designed specifically for pest management.

Public Education

The most effective measure for preventing injury to park visitors and personnel is education. Visitor activities should be directed away from highly infested areas. Park visitors should be informed about the habits of fire ants, how to recognize them, and how to avoid them. Visitors should be encouraged to use proper sanitation so that fire ants are not attracted to such sites as picnic areas. And if the worst happens, information should be available on what to do if a person is stung.

CHEMICAL CONTROL OF FIRE ANTS

Many different types of chemical products are available for fire ant control. There are three major ways to manage fire ants with chemicals: treating individual mounds, broadcast treatment of a large area, and spot treatment in and around structures. Remember to consult with your IPM coordinator for specific pesticide recommendations for your area.

Mound Treatment

Treating individual ant mounds is time consuming, but it is generally the most effective method of control. It takes from a few hours to a few weeks to "kill" the mound, depending on the product used. Individual mound treatment is usually most effective in the spring. The key is to locate and treat all the mounds in the area to be protected, not always a simple task. If many young mounds are missed, reinfestation of the area can take place in less than a year. The following discussions describe different ways to treat individual mounds.

Mound drench. Follow directions for dilution of the insecticide and gently wet the mound and surrounding area with insecticide. Then break open the top of the mound and pour the insecticide dilution directly into the galleries.

Mound drenches are most effective after rains when the ground is wet and the ants have moved up into the drier soil in the mound. During excessively dry weather, effectiveness of the treatment may be enhanced if you soak the soil around the mound with plain water before you treat.

A few granular insecticides are labeled for application to fire ant mounds. After application, the granules are watered into the mound.

Mound injection. A growing number of insecticide products are designed to be injected directly into fire ant mounds. They may be injected using a "termite rig" with a soil injector tip, a standard 1-3 gallon compressed air sprayer with a fire ant injector tip, or a special aerosol soil injector system. The mound is injected in a circular pattern, usually at 3 to 10 points. A new product combines insecticide treatment with high temperature vapors to increase penetration in the mound.

Baits. A few bait products are available that may be used for individual mound treatments. The baits take from several days to several weeks to eliminate a fire ant colony, but they can be very effective and are simple to use. Baits are available with the either a toxicant, a sterilant/toxicant, or a growth regulator. The baits are sprinkled around and sometimes on the mounds. During hot weather it is best to apply the bait late in the afternoon or early in the evening when the ants begin to forage. Baits must be kept dry.

Dusts. A few insecticide dusts are labeled for dusting individual fire ant mounds. The dusts are evenly distributed over the top of the mound. Dusts must be dry in order to be effective.

Fumigation. Large fire ant mounds can be eliminated through fumigation. Check with your IPM coordinator to see if these products are registered for use in your area. Only those who have been specifically trained in the use of fumigants should conduct such fumigations.

Broadcast Treatment

Several different types of products are labeled for application over wide areas to control fire ants. Granular insecticides are often applied with hand-operated fertilizer spreaders or agricultural application equipment. Sprays also are sometimes used. Because of the broad spectrum of such treatments and their effects on nontarget species, broadcast application of standard insecticides is not a good choice for park land.

A better choice is broadcast treatment with an insect growth regulator bait, which poses much less risks to nontarget species. For example, fenoxycarb bait has been shown to be very effective for suppression of fire ant populations when applied in one application over a wide area.

Spot Treatment with Insecticides

If fire ants are nesting in a structure (in a wall void, for example), the nest should be treated directly, usually by drilling and injecting with a residual insecticide. Treatment of ant trails or barrier treatment to keep fire ants from foraging in occupied areas are generally not acceptable choices for Park Service facilities.

Summary of Management Recommendations

Park visitors and personnel should be directed away from infested areas and encouraged to observe proper sanitation procedures so that fire ants are not attracted to recreational sites.

Mechanical and other nonchemical control measures should be considered first if control is deemed necessary. Remember that control may not be necessary in many cases. When it is necessary, chemical control, particularly the use of baits, may be attempted if other control measures have failed and the use of pesticides has been approved by the National Park Service.

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Fleas

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for fleas. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Although there are over 250 species of fleas described in North America (Pratt 1957), only a few are commonly encountered by humans with enough frequency to be considered pests (Ehman and Story 1982). These include the cat flea, *Ctenocephalides felis* (Bouche), the dog flea, *C. canis* (Curtis), the human flea, *Pulex irritans* (L), and the oriental rat flea, *Xenopsylla cheopis* (Rothschild). Other species, such as the rabbit flea, *Cediopsylla symplex*, the mouse flea, *Ctenopsyllus segnis*, the ground squirrel flea *Diamanus montanus* (Baker), and *Oropsylla hirsuta*, a flea that feeds on prairie dogs, may achieve pest status when their host mammals nest in or near structures or the fleas attack hunters and hikers. Some, such as the northern rat flea, ground squirrel flea, and *Oropsylla hirsuta* are important vectors of sylvatic plague, bubonic plague, and murine typhus.

Flea management is best done via management of the host animal's habitat. Since fleas must spend part of their life cycle on their host, the chances of encountering fleas in areas of the host's habitat where it spends most of its time (e.g., its den or nest) are much greater than in a general area, such as a field or barn in which the host may or may not be found at a given time. One author has suggested that most fleas spend more time in the host nest or burrow than on the host itself (Benton 1980). This is the emphasis that will be placed on flea management strategies in this module.

IDENTIFICATION AND BIOLOGY OF FLEAS

This document will deal with the four most commonly encountered flea species mentioned above: the cat flea, the dog flea, the human flea, and the oriental rat flea. These fleas are found throughout the United States and are most likely to be encountered in mammal and bird nests or in pet bedding. Adult fleas are ectoparasites of their hosts, but unlike many other ectoparasites they do not spend the majority of their life cycle on their host.

Females deposit eggs in groups of 1 to 18 on the host after a blood meal. Some species, such as the cat flea, can deposit up to 25 eggs per day and over 1000 in a lifetime. Eggs soon drop off or are brushed off. Due to their spherical or oval shape, they roll into cracks and crevices on the floor or in or near nests and bedding. Eggs are whitish and 1/32" in diameter. Eggs hatch in 2 to

21 days.

Larvae are approximately 1/4" when first hatched, white, and have fine hairs. They lack legs or eyes but possess biting mouthparts. Most species feed on dried blood from the host (in the form of adult flea feces) or organic debris present in cracks and crevices. They also feed on cast larval skins. Depending on the availability of food, relative humidity, and other environmental factors, larvae pass through three stages (instars) in one week to several months. Optimal temperatures for larval development are 65 to 80F. Larvae need a relative humidity of at least 50%. It is important to realize that even if the relative humidity of the ambient air is not this high, it could be much higher in the microhabitat of a burrow or den. Larvae can also survive short exposures to below freezing temperatures (Silverman and Rust 1983). Larvae pupate within cocoons spun from silk and may be covered with debris.

The pupal stage usually lasts approximately one week. The newly emerged adult may remain in the cocoon for some time; under adverse conditions, the adult may spend up to a year in the cocoon. Emergence occurs in response to pressure applied to the cocoon or detection of host warmth, vibrations, or carbon dioxide in the host's breath.

Adult fleas are small, brownish insects flattened from side to side, without wings but with powerful jumping legs. Adults can live for several years and go without feeding for months at a time under extreme conditions. Fleas can remain in a structure long after the host mammals have been removed. Depending on the species and environmental conditions, adults can breed from two weeks to two years after emerging. Adults feed on blood, and females deposit eggs only after a blood meal. Most species remain on the host only long enough to feed. Nearly all species have host preferences but are not restricted to any one host species. This trait is responsible for the transmission of several diseases (e.g. plague or murine typhus) from one host species to another. Adults prefer warm humid places and will leave a host if it dies.

Outdoors, fleas are most abundant during humid, rainy summers and are more common outside in the southern United States than in the north. Indoors, warmth and high relative humidities are conducive to large populations. The sudden appearance of large numbers of adult fleas in mid-summer and fall ("flea seasons") is due in large part to the onset of higher humidities and temperatures which permit larval development to accelerate. Larvae may undergo arrested development in less than favorable conditions.

Medical Importance of Fleas

Flea bites vary in effect from short-lived itching welts to an overall rash to symptoms which may last over a year, depending on the sensitivity of the victim. Young children are more sensitive than older persons. Commonly, a small red spot appears where the skin has been pierced. Little swelling ensues, but the spot is accompanied by a red halo of irritated skin which usually lasts for several hours to a day.

Fleas are vectors of several diseases important to human health including plague, murine typhus, and tularemia. The oriental rat flea is the most important plague vector from rodents (primarily rats) to humans, but at least 30 other flea species can also transmit the disease, including the northern rat flea, dog flea, cat flea, and the human flea. Plague (in the sylvatic form) is endemic in the western United States among small rodents such as chipmunks, ground squirrels, and prairie dogs.

Nearly all known cases of plague in humans in the United States since 1925 have been associated with wild rodents (mostly from the Rocky Mountain states) and their fleas. The greatest threat to humans exist when domestic rats are exposed to infection from wild rodents in areas adjacent to human communities.

Murine typhus is a mild form of epidemic typhus that is usually spread by the human louse. The Norway rat population is the main reservoir of the disease. The disease is most common in the southwestern and Gulf states. The disease is commonly spread from rat to rat, and from rat to human by the oriental and northern rat fleas. It has also been transmitted by cat fleas from infected feral cats.

Fleas are also vectors of tularemia, a disease related to plague. The natural reservoirs of tularemia are cottontail rabbits in the East, and jack rabbits in the West. Most cases reported are by hunters.

Fleas can also be intermediate hosts of several species of tapeworm including species which parasitize humans, dogs, and cats.

MONITORING AND THRESHOLDS FOR FLEAS

Fleas can be monitored in several ways. The simplest is to count and collect fleas landing or crawling on an observer's lower legs for one minute. In making surveys, trousers should be tucked into white socks to prevent bites and make collecting easier (socks can be put on over shoes). Light-colored trousers are preferred to provide greater contrast and facilitate counting and collection. A variation on the above is to wrap fly paper (sticky side out) around the lower legs and count fleas adhering after a predetermined interval (Cole and Burden 1978).

Fleas may also be combed off animals for an index of animal infestation. Do this over a white surface so fleas can be easily observed (Ehmann and Storey 1982).

Pet bedding should be periodically checked for flea eggs and dried-blood feces (frass) of adult fleas. This has been described as "salt and pepper" because it looks like small flecks of black and white debris. The frass is generally cylindrical, twisted, and about 1/16" long. It is dark in color. Larvae and pupae can be found at the edge of pet bedding or animal nests.

Indoors, five or more fleas on the legs of observers in less than one minute is indicative of severe infestation.

Flea populations in animal burrows or dens can be sampled by using a flannel cloth that is run through the burrow on the end of a plumber's snake. The number of fleas on the cloth is then counted. See Barnes et al. (1972) for more detail.

NON-CHEMICAL CONTROL OF FLEAS

Several studies have indicated that fleas spend the majority of their life either on the host or in the host's bedding or nest, so flea management should focus on these. In outdoor settings, the emphasis should be on spot treatment of nests with an insecticide. Exclusion of the host animal from an area may be desirable as well, but the feasibility of this strategy will vary with the animal and the location of its nest. In the case of domestic animals, sanitation should be the focus of a flea management program. Regular cleaning of bedding and other areas where the animal spends the majority of its time should reduce flea populations to non-irritating levels.

In areas where plague is endemic (e.g. the southwestern United States), efforts should be made to keep humans and fleas (and their wildlife hosts) separate. Prairie dog towns should not be allowed to expand into campgrounds and other developed areas. Camping and other outdoor activities should be restricted during an outbreak when fleas seek other hosts. Prairie dog burrows can be dusted with insecticide. Check with Public Health Service officials if your area is affected.

In most other cases, fleas are considered pests due to the nuisance caused by their bites. In these situations, management decisions should be made on a case-by- case basis.

Sanitation

Fleas require warm-blooded hosts for development and for egg maturation. Elimination of suitable habitat for wild rodents and other animals near structures will often reduce flea population levels. Screened vents prevent animals from resting inside or underneath structures. Eliminating vegetation close to structures and raising woodpiles off the ground reduces rodent harborage.

Indoors, wash or vacuum all pet bedding and sleeping areas on a regular basis. Cracks and crevices should be vacuumed and sealed, especially the area between the baseboard and floor. Dispose of vacuum cleaner bags to prevent reinfestation. Pets should be washed regularly and treated with insecticides if necessary.

Ultrasound

The ultrasonic collar is sometimes for the control of fleas on domestic animals. A recent study (Hinkel, Koehler, and Patterson, 1990) showed that ultrasound devices are ineffective.

Insect Growth Regulators

A new technology in the management of fleas is the use of insect growth regulators (IGRs). These substances are similar to chemicals produced by the flea to regulate the shedding of its skin during molting. They work by interfering with the molting process, thus preventing the immature flea from developing into an adult. This method of control is a long-term process, since it will only kill larvae as they molt. A recent study using pyriproxyfen (sold as Nylar), an insect growth regulator reported to be effective against several insects, examined its effectiveness against the cat flea. One problem with insect growth regulators is that they break down when exposed to light, limiting their outdoor use. In this study, Nylar was determined to be stable when exposed to light. It was found to persist in home yards for three weeks after application and to prevent development of 90% of the fleas in treated areas (Palma and Meola 1990). Another effective IGR for flea management indoors is methoprene (trade name Precor). It is important to combine the use of a material such as this with observations of the infested animal's movement so that only areas where it spends the majority of its time are treated.

Flea Predators

Fleas are preyed upon by ants and beetles that feed on larvae in the host's nest (Fox and Bayona 1968).

CHEMICAL CONTROL OF FLEAS

Insecticides are also part of a flea management program. These are applied to areas where fleas are most likely to breed, including animal bedding, cracks in floors, and baseboards. Many veterinarians also recommend the use of indoor foggers to apply pesticides to rooms where domestic animals spend the bulk of their time. Flea collars are not considered to be effective (Whiteley 1987). When insecticides are used, it should be in conjunction with sanitation (Arthur et al. 1989). One difficulty with the use of insecticides as part of a flea management program is the ability of the adult flea to remain in its cocoon as a preemerged adult. This means that the adult flea can remain in the cocoon in which it pupates until it encounters a suitable host. Insecticides have been found to be ineffective against these preemerged adults (Rust and Reierson 1989). This highlights the importance of sanitation as the key element in a flea management program.

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Gypsy Moth

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for gypsy moth. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

BIOLOGY AND IDENTIFICATION OF THE GYPSY MOTH

Lymantria dispar (L). The adult female moth is dirty-to- creamy white, with dark bands across the forewings. Adult females have a wingspan of about 2" but can only fly short distances. The female's body is stout and densely covered with hairs, and her antennae appear thread-like. The male is much darker and smaller than the female; the wings are dark brown with black bands across the forewings. The wingspan is about 1/4", and the antennae are feathery. The abdomen is narrower than the female's.

Eggs are globular, whitish, and about 1/32" in diameter. They are laid in oval masses of 75-1,000 (averaging 400-500) and are covered with buff-colored hairs from the female's abdomen. Egg masses may be 1/2"-2" long, depending on their shape.

Newly-hatched larvae are buff colored but turn black within four hours after emergence. Younger caterpillars (first to fourth instars) are brown to black in color with long body hairs. Later instars are black with 11 pairs of colored tubercles, or bumps, along the top surface. The front five pairs are blue, and the rear six pairs are red. Each tubercle is topped by a tuft of yellow or brown hairs, which may be up to half a body-length long. A yellow line runs along the top surface from the head to the last body segment. In the fourth through sixth instars, the dark-colored head has additional yellow lines. The true legs are dark red.

Pupae are teardrop-shaped, chocolate to dark red-brown in color, and rounded in the front and tapered at the rear. Male pupae are 3/4"-1/2" long, while female pupae may be up to 2 1/2" long. A few hairs may occur on the head and each abdominal segment. Each pupa is attached to the substrate by a few strands of silk.

See McManus and Zerillo (1978) for a photographic guide to all life stages of the gypsy moth.

Geographic Distribution

The gypsy moth is an exotic species that was accidentally introduced into Massachusetts in 1869.

Since then, it has spread west to Ohio, south to North Carolina, and north to Montreal, Canada. An isolated population has also become established in central Michigan. Scattered infestations occur from time to time in states outside of the generally-infested area described above, but usually these are quickly detected and eradicated.

A recent analysis (Liebhold et al. 1991) indicated that the spread of the area generally infested by the gypsy moth ranged from 1/2-3 miles/year during 1900-1965. Since 1965, the rate of spread has been 3 1/2 miles/year in areas with a January mean temperature of less than 44F, and 10 miles/year in areas with a January mean temperature greater than this. Based on this analysis, the gypsy moth is expected to spread west to Wisconsin and Iowa and south to Georgia by the year 2015.

Habitat

Temperate and boreal deciduous forests are the favored habitats of the gypsy moth. Defoliation occurs most frequently in forests on dry ridges and steep slopes that have shallow soils, and on sandy plains that have deep, excessively-drained soils. Outbreaks also occur frequently along interfaces between forests and urban areas (Houston and Valentine 1985). High population densities (or transport as a result of human activities) may result in migration to nearby or distant softwood forest, urban, or agricultural environments, all of which may support gypsy moth populations on available plant foliage.

Hosts

The leaves of close to 500 species of trees and other plants can be eaten by gypsy moth larvae (adults do not have fully- developed mouthparts and therefore do not feed). Table 1 provides some information on host preference based on laboratory and field observations.

Actually, host preference and suitability is more complicated than implied by Table 1, as some tree species are not suitable hosts for first instars gypsy moths, but are good hosts for later instars. This is the case for many coniferous tree species. Also, some hosts are suitable for only a short period of time, after which they undergo physiological changes that reduce their suitability. For instance, beech is suitable for young larvae for less than one week (Raupp et al. 1988).

Gypsy moths attack trees under stress (e.g., from drought or natural or man-made disturbance) more readily than healthy trees. The presence of bark flaps and deep-bark fissures, which provide hiding places for larvae, are considered important in determining susceptibility of forest stands to gypsy moths.

Table 1. Host Plant Preferences of the Gypsy Moth Relative to Red Oak. (Modified from Montgomery and Wallner [1988]).

	Laboratory rearing[a]	Defoliation level[b]	Defoliation level[cd]
Red Oak	++	1.00	1.00
Black oak	++	1.13	1.35
Chestnut oak	++	1.11	1.47
White oak	++	1.11	0.83
Aspen	++	1.18	1.10
Basswood	++	1.24	0.56

White birch	++	0.76	0.56
American beech	++	0.50	0.54
Red maple	+	0.42	0.42
Sugar maple	+	0.20	0.68
Hickory	+	0.33	0.76
Black cherry	+	0.44	0.29
White pine	+	0.34	0.22
Hemlock	+	0.24	0.01
Black locust	- -	0.20	0.15
Ash	- -	0.07	0.20

[a] ++, favored host; +, acceptable; --, avoided; Massachusetts (Mosher 1915).
[b] 190 plots surveyed for 20 or 30 years in New England (Campbell and Sloan 1977).
[c] 575 plots surveyed I year in Pennsylvania (Gansner and Herrick 1985).
[d] values are the ratio of average defoliation of the indicated species to average defoliation of red oak.

Life Cycle

Gypsy moths produce only one generation each year. Adults emerge from pupae during June in the southern parts of their range and July-August in the northern parts of its range. Emergence is accelerated under extremely high-density conditions. Males usually appear one to two days prior to females and fly in zig-zag or (less commonly) straight patterns. Vertical objects such as tree trunks where females are most likely to be found attract the males. Most males will fly less than 1/2 mile (usually fewer than 650') from their site of emergence. Females do not fly.

Several hours after emerging, females release a sex pheromone in bursts from abdominal glands. This chemical attracts males, who follow the scent upwind to locate the female and begin mating. Mating may last up to 1/2 hour, and females begin depositing eggs within 24 hours (Giebultowicz et al. 1991). Multiple mating may be common among males, but is probably rare among females, since the release of pheromone is inhibited by mating. Adult moths live about one week.

Generally, gypsy moths lay their egg masses are found on tree trunks and the undersides of branches, in crevices, under loose bark, and under or on rocks, tree stumps, foliage, or vehicles. The egg stage lasts for eight to nine months. Hairs from the female's abdomen surround the eggs, providing some protection from winter temperatures and natural enemies. Larval development is completed inside the eggs about a month after laying, but the larvae enter diapause and do not emerge until the following spring. Egg hatch usually begins at about the same time that red oak buds open.

Most larvae will hatch from an egg mass within a week, but the hatch period may be up to a month in egg masses in cool, shaded, or high-altitude areas. Newly-hatched larvae are about 1/8" long and remain near their egg mass if the weather is rainy or if temperatures are below 45^{o}F. Once they have left the egg masses, larvae are attracted to light and move upwards, spinning a thread of silk, until they reach the top of the tree or other object on which they hatched. Under some conditions, they may spin down on silk threads. If the wind is strong enough, the threads may break and carry the larvae up to 650' within the forest canopy. Rarely, larvae may disperse up to 12 miles if they are carried out of the canopy by updrafts (Montgomery and Wallner 1988, Taylor and Reling 1986).

Larvae feed first on new leaves. When not feeding, the young larvae stay on the undersides of leaves, where they form a silk mat on the leaf surface for attachment. Molting occurs at intervals of about one week, which allows the larvae to grow in size. Males usually undergo four molts and females usually undergo five, but as many as nine have been recorded. After the third molt, when population density is low to moderate, larval behavior changes dramatically. Rather than remain always in the canopy, larvae leave the foliage during daylight hours and seek hiding places on the boles of trees or on the ground. Under high-density conditions, even large larvae remain in the canopy during the day.

At the end of the larval period, each larva seeks a pupation site, surrounds itself with a sparse silk net, rests for one to two days, and then becomes a pupa. The pupa breaks out of the larval cuticle, turns dark brown, and remains in its silk net for about two weeks. When development is complete, the newly-formed adult breaks out of the pupal skin, expands its wings over a period of several hours, and begins its adult life.

Population Cycles

Gypsy moth populations exist in four distinct phases (Elkinton and Liebhold 1990). The **innocuous** phase is characterized by very low population levels. Gypsy moth life stages are often difficult to find during this phase, which may persist for several years. The **release** phase usually takes place over one to two years and can result in population density increases of several orders of magnitude. The **outbreak** phase is characterized by populations high enough to cause noticeable tree defoliation. Outbreaks are rarely sustained for more than one to two years, after which high levels of mortality, primarily from starvation and disease, bring about a rapid population crash. This is the **decline** phase. These population changes often occur synchronously over wide geographical regions. However, there is little evidence that gypsy moth population outbreaks occur in regularly spaced cycles in North America (Elkinton and Liebhold 1990).

Responses to Environmental Factors

Temperature: Exposure of eggs to temperatures of less than - 45°F causes high mortality. Exposure of larvae to freezing temperatures may be lethal. Larval development is accelerated up to one to two weeks under outbreak conditions, probably as a result of behavioral changes which lead to greater exposure to higher temperatures (Elkinton and Liebhold 1990).

Moisture: Heavy rainfall at hatch may result in drowning of larvae. Rainy weather during the first larval instar can delay migration and cause larvae to congregate on the undersides of leaves. The duration of this instar may increase under these conditions. Extended congregation may stress larvae and increase their susceptibility to nucleopolyhedrosis virus (also known as "wilt"). Rainfall and moisture appear to increase the transmission of the gypsy moth fungus *Entomophaga maimaiga* (Weseloh and Andreadis 1992).

Light: Gypsy moth larvae are attracted to light just after hatch, leading them to move upward to sites from which they can be transported by wind (McManus 1973). Young larvae (instars one through three) remain on foliage during the day, while older larvae alter this behavior, resting away from the canopy during the day and returning to feed at night. Adult emergence and male sperm release are also triggered by daily light/dark cycles (Giebultowicz et al. 1990).

Wind: Larvae disperse mainly by wind. Newly-hatched larvae trail silk as they climb to treetops or the upper surface of the objects on which they hatch. These larvae are most active during the daytime, when winds are usually strongest. When they encounter wind, they arch their bodies (to catch the wind) and extrude a silk thread which may act as a balloon or parachute. In addition, first instar larvae are covered with comparatively long hairs, which increase their buoyancy in air.

Foliage Chemistry

Gypsy moth feeding has been shown to decrease the nutritional value and increase the levels of toxic chemicals in the remaining foliage (Montgomery and Wallner 1988). As a result, larvae grow more slowly and gain less weight on defoliated oak trees. In some cases, these changes may contribute to the decline of high populations (Schultz and Baldwin 1982).

Population Density

Under outbreak conditions, development time is reduced (by up to two weeks), sex ratios become male-biased, and smaller adults which lay fewer eggs are produced (Elkinton and Liebhold 1990). Older larvae at innocuous population densities feed in the canopy only at night and seek protected resting places during the day. Under outbreak conditions, late instars remain in the canopy and feed intermittently throughout the day and night; however, they appear to consume no more foliage than larvae from innocuous densities.

Impact of the Gypsy Moth

The gypsy moth is one of the most destructive defoliators of hard and softwood trees. Tree mortality resulting from gypsy moth defoliation is highly dependent on the interaction between tree species, tree health, environmental stresses, and the severity of defoliation. Mortality to overstory oak trees subjected to gypsy moth defoliation in Pennsylvania ranged from 13% for trees with good initial crown condition to 35% with poor initial crown condition (Herrick and Gansner 1987). Mortality averaged 67% for understory oaks. Over 84% mortality of white oaks following defoliation was recorded in a New Jersey forest (Kegg 1973). In addition to aesthetic problems and reductions of timber stand value due to defoliation, forests suffering gypsy moth attack may suffer increased risks of fires due to canopy reduction and accelerated drying of litter. Effects of defoliation on watershed output and water quality are unclear at present (Corbett and Lynch 1987). In recreation areas, unsightly defoliated areas and wandering larvae can result in decreased visitor use and revenues (Goebl 1987).

Defoliation of forest trees can lead to increased susceptibility to other pest damage, most frequently invasion by the shoestring fungus, *Armillaria mellea*, and the twolined chestnut borer, *Agrilus bilineatus*, and alteration of ecological succession at affected sites (Houston 1981). The long-term effects of tree defoliation and mortality on the forest ecosystem are not known.

Asian Gypsy Moth

The currently established North American population of gypsy moths was introduced into Massachusetts from France in 1869. Until recently, there was no evidence of subsequent introductions. In 1991, gypsy moth egg masses on a Soviet ship docked in Vancouver, British Columbia, were found to be hatching. Because it was feared that larvae may have blown onshore, steps were taken to detect and identify new gypsy moth introductions into northwestern North America. During the summer and fall of 1991, asian gypsy moth adults were found in Portland, OR, and Tacoma, WA, in the United States, and in Vancouver, British Columbia in Canada. A mitochondrial DNA sequencing technique is presently used to distinguish the asian gypsy moths from the North American gypsy moths. Eradication efforts and extensive delimitation trapping programs were initiated in 1991 in all three of these locations.

The asian gypsy moth is similar in appearance to the North American gypsy moth, except that the asian larvae vary more in color. Asian females, unlike flightless North American females, are strong fliers (>20 miles). Lights attract asian females, and they lay their eggs on foliage and on objects near lights, in addition to tree trunks and other objects. In its native range, the asian gypsy moth feeds on at least 600 plant species and appears to thrive better on marginal hosts than the North American gypsy moth.

MONITORING AND THRESHOLDS FOR GYPSY MOTHS

Population Monitoring

Several methods are available for monitoring gypsy moth populations. The choice of method should be based on the population level suspected, location of sampling site in relation to the established United States infestation area, and resources available. The U.S. Forest Service currently provides gypsy moth survey assistance to any federal agency on request, and should be consulted if you wish to have a survey conducted.

Adult male trapping: These techniques involve the use of special traps baited with a synthetic form of the sex pheromone produced by receptive female gypsy moths. The trap currently used for gypsy moth surveys by the U.S. Forest Service and the USDA Animal and Plant Health Inspection Service (APHIS) are fully described by Schwalbe (1979). Although several variations of the trap design are manufactured, the USDA-approved traps can be obtained from your regional U.S. Forest Service office.

Pheromone traps should be placed before male moths begin flying (see Life Cycle section, above). Schwalbe (1979) describes the use of pheromone traps to detect low gypsy moth populations (detection survey) and to define specific areas of infestation (delimiting survey). An effective technique only for relatively low populations, pheromone trapping is recommended for use in areas outside (or on the edges of) established infestations.

The interpretation of pheromone trapping results is subjective; no reliable relationships between numbers of trapped males and gypsy moth population density have yet been found. Currently, for detection surveys, APHIS recommends placing pheromone traps at a density of one trap per 1 to 4 square miles and at frequencies of every two or four years, depending on the potential for accidental introductions to occur in a particular area (Anonymous 1990). When moths are captured during a detection survey, a delimiting survey may be conducted in the vicinity of the trap catches. In delimiting surveys, traps are deployed at densities of 16-36 traps per square mile over areas of from 1 to 4 square miles. The pattern of trap catches can be used to estimate the approximate area of infestation.

Larval trapping: The collection of gypsy moth larvae under burlap bands, while not useful in quantifying population density, can serve as another early indicator of low (e.g., recently established) but building populations. The most convenient method involves tying a 12"-wide burlap band around the trunk of each tree to be monitored so that the top 6" of the band can be pulled down over the bottom, making a shaded flap in which larvae will hide during daylight hours. Bands should be monitored two times each week and any trapped larvae should be destroyed. The presence of gypsy moth larvae in such traps indicates that a population may be developing in the vicinity of the trap site and that other survey methods should be used to determine whether treatment is required. Tar-paper wrappings and plastic tree flaps can be used instead of burlap.

Egg mass counting: Several methods have been developed for determining the number of gypsy moth egg masses in an infested area. Egg mass counts can be done from the time of oviposition (usually June-August) until egg hatch the following April or May. Counts are easier and probably more accurate, after the leaves have dropped from deciduous trees. The walks generally follow an "M"-shaped pattern through the area to be sampled, which helps to eliminate an edge effect. In forest situations, edge trees have found to have 2.4 times more egg masses than interior trees (Bellinger et al. 1989). Methods currently in use include:

Threshold walk: An observer walks through the area to be monitored, counting all new (current season) egg masses. The walk ends when the count reaches a predetermined number (see Threshold/Action population levels, below). This method gives no approximation of the actual gypsy moth density in an area, but it is easily done, and in areas of high gypsy moth density it may be useful in making a treat/no-treat decision using accepted threshold values.

Five-minute walk: Two observers walk through the area to be monitored for a five-minute period; each counts every detectable new egg mass. The average of the two counts is calculated and converted to an approximate number of egg masses per acre by the following equation:

estimated number of egg masses per acre = (average number of egg masses observed x 20) + 15

(Schneeberger 1987). The estimated number of egg masses/acre can be compared to established threshold levels to determine whether treatment is necessary. A recent analysis of this method (Liebhold et al. 1991) recommends against its use because density estimates vary too much among observers and because it is generally too imprecise.

Intensive search. This method is used for very small populations (i.e., no evident defoliation, but with multiple adult male catches in pheromone traps) and simply involves examination for egg masses on **all** surfaces in the vicinity of traps with trapped males, including under bark flaps, on rocks, and in tree holes. It can be quantified somewhat by reporting the number of egg masses found per person-hour of searching. Intensive searching is recommended to support pheromone trapping for the discovery of new infestations.

The following two methods are currently the only methods available for quantifying gypsy moth egg mass density. Both methods consist of a complete census of all egg masses occurring within a predetermined number of randomly located sample plots. The number of egg masses per acre is estimated from the samples.

Fixed-and variable-radius plot counts. This method is described in detail by Wilson and Fontaine (1978). At each sample plot, all of the egg masses are counted on trees selected from the plot center using a prism (a tool commonly used by foresters for estimating tree basal area). Egg masses occurring on the ground are counted within a fixed-radius plot located around the same plot center.

Fixed-radius plot. The observer counts every new egg mass on trees and on the ground within a circle with a radius of 18.6 feet (1/40th acre or 0.01 ha) around a chosen point. This count multiplied by 40 gives an estimate of the number of egg masses per acre. This method is more cost effective than the fixed- and variable-radius plot method (Kolodny-Hirsch 1986; Thorpe and Ridgway 1992) and is currently the most widely used method.

In addition to providing an estimate of the number of egg masses in an infested area, these methods can provide the opportunity for the observer to judge the health of the gypsy moth population. Egg masses that are thick and of large size (about that of a 50-cent piece), showing little or no parasitoid damage (such as small holes) and containing large quantities of undamaged fertile eggs indicate a healthy population. In many cases, a numerically large population of small egg masses or those showing predator/parasitoid injury may indicate a declining gypsy moth population which may not require treatment. Unfortunately, assessment of gypsy moth population quality must be done subjectively, as analytical guidelines do not exist.

Sequential sampling plans. A sequential sampling plan for gypsy moth has been used successfully in Shenandoah National Park as well as in several urban areas in northern Virginia for five years (Ravlin 1991; Ravlin 1994). Sequential sampling is a process in which a given number of samples are taken and, based on how far above or below the threshold you are at the end of the sample, a decision is made to either stop sampling and apply a treatment, stop sampling without a treatment, or continue sampling to gather more information before a decision about gypsy moth management is made.

Of course, any egg masses found in areas outside the established North American infestation area may represent the spread of the gypsy moth and may require treatment, since isolated infestations can usually be eradicated. Within the infested region, management of the moth population to limit defoliation and population growth is the most sound approach, since eradication is impossible.

Defoliation Monitoring

In addition to directly sampling the gypsy moth population in a particular area, site managers may wish to indirectly track zones of defoliation to determine where to treat otherwise unidentified populations, where to set up traps next spring, and the spread of existing infestations. Defoliation should also be monitored to assess the efficacy of any treatments that were applied. Defoliation is generally monitored during the period of peak larval development in one or more of the following ways.

Ground estimation: An observer may make estimates of percentage defoliation of particular trees by walking through the infested area and examining tree crowns through binoculars. A slightly more comprehensive method involves using the fixed- radius or fixed- and variable-radius plot designs noted above (under Egg Mass Counting), and again estimating the percentage defoliation noted on each tree observed. Comparing photographs of a sample area taken at regular intervals will allow the observation of changes in canopy density due to defoliation. These methods are very time-consuming and are subject to errors of interpretation. They are discussed by Talerico (1981).

Sketch mapping: An observer may fly over the area to be monitored in an aircraft, sketching zones of light, medium, or heavy infestation on a U.S. Geological Survey map of the area. Talerico (1981) details the procedure and interpretation of such maps. As in ground observation methods, interpretation of the results is largely a matter of experience.

Threshold/Action Population Levels

To date, efforts to construct reliable predictive models for gypsy moth defoliation based on population density have been only partly successful at best (Ganser et al. 1985, Montgomery 1990). Current defoliation thresholds are rough estimates (Figure 1). The following population values are currently used by the U.S. Forest Service and APHIS in their gypsy moth management programs. The National Park Service follows U.S. Forest Service guidelines. It should be noted that the goal of the U.S. Forest Service Forest Pest Management program differs depending on whether or not the gypsy moth population is within the area of the United States that is recognized as being generally infested. Contact your regional U.S. Forest Service office or regional Integrated Pest Management coordinator for this information.

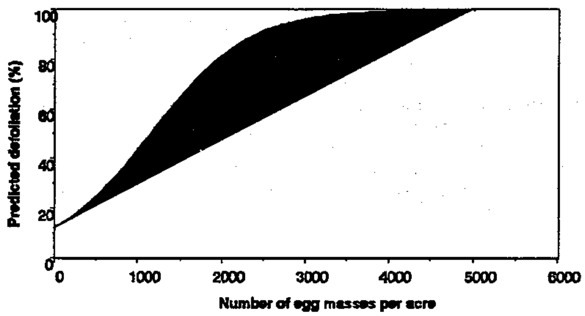

Figure 1. Predicting defoliation from the number of egg masses per acre (from Gansner et al. 1985).

For areas in established infestation zones: At 250-500 egg masses/acre, gypsy moth populations may produce noticeable defoliation. Treatment is recommended for high-use recreational areas (campgrounds, trailer parks, and other areas with transient traffic) and residential areas.

For areas outside established infestations: The capture of any male moths in a detection survey may indicate the need to conduct a more intensive delimiting survey. The decision to delimit the following season should be based on such factors as history, host vegetation, local resources, and movement of people in the vicinity (Anonymous 1990). If the delimiting survey indicates that an isolated population has developed, further delimiting surveys, intensive searches for egg masses, and eradication treatments may be indicated.

NON-CHEMICAL CONTROL OF GYPSY MOTHS

Individual Tree Treatments

Egg mass destruction: Scraping and removing egg masses is one of the oldest methods used against the gypsy moth in North America. In residential areas, where 50% of the egg masses may occur within reach of the ground (Thorpe and Ridgway 1992), this approach could destroy a significant portion of the population. However, because of the tendency of larvae to migrate in from adjacent areas, scraping should not be relied upon for effective control. Vegetable oils have been shown to be effective ovicides when applied to egg masses in the fall (Ralph Webb, in manuscript), and a soybean oil product is registered for use on gypsy moth egg masses.

Barrier bands: Sticky barrier bands placed on tree trunks can prevent larvae from crossing (Webb and Boyd 1983); there is some evidence that under outbreak conditions and on isolated oak trees, barrier bands can reduce defoliation can be reduced (Blumenthal 1983). However, under gypsy moth population densities capable of causing less than 60% defoliation, larval populations on banded trees are reduced only an average of about 25%, and defoliation reduction is highly variable (Thorpe et al. 1993). Sticky barrier bands are available commercially or can be made from duct tape and Tree Tanglefoot. Tanglefoot should be applied to the tape and not the surface of the tree because it can damage bark. Since they can reduce larval populations somewhat, and because of their low cost, sticky barrier band use may be advisable on high value, individual trees when no other treatment will be used. However, sticky barrier bands alone should never be relied upon to prevent defoliation.

Burlap bands can be used as a control tactic if they are checked frequently and all larvae resting beneath them are destroyed. The efficacy of this method has not been quantitatively evaluated.

Natural Enemies of Gypsy Moth

Naturally occurring predators and parasitoids of the gypsy moth, while numerous and abundant, are not capable of preventing outbreaks. Efforts to control gypsy moths by rearing and releasing large numbers of parasitoids have not been successful (Blumenthal et al. 1979, Kolodny-Hirsch et al. 1988). The best way for a site manager to make use of available natural enemies of the gypsy moth is to use management alternatives (e.g., *B.t.* or no treatment) which will not adversely affect the natural enemies, leaving them to function as a part of a gypsy moth integrated pest management program. See Blumenthal et al. (1981) for a detailed discussion of predator/parasitoid research. Egg mass surveys and larval surveys can include observations of predator/parasitoid presence as a guide to maximizing their effectiveness.

Pathogens

Bacteria: The naturally-occurring bacteria *Streptococcus faecalis* and *Pseudomonas* spp. occasionally cause high levels of mortality (up to 60%) under outbreak conditions (Podgwaite 1981).

Nucleopolyhedrosis virus: A virus of the genus *Baculovirus* is closely associated with all North American gypsy moth populations. Its effects are most often seen under outbreak conditions, when a large proportion of the larval population may be killed. For more information on this disease, see the following section on area-wide suppression of the gypsy moth.

Entomophaga fungus: For the first time in 1989, the fungal disease *Entomophaga maimaiga* was reported causing widespread mortality to North American gypsy moth populations (Hajek and Soper 1992). This disease was known to cause extensive mortality in Japan. It is now known to occur in 13 states from Maine to Virginia (Elkinton et al. 1991). The appearance of larvae killed by *Entomophaga* is similar to that of virus-killed larvae, and definitive identification requires examination by an expert.

Parasitoids

Since 1905, more than 40 species of parasitic flies and wasps have been introduced into North America to control the gypsy moth. Among the 10 which have become established are the egg parasitoids *Ooencyrtus kuvanae* and *Anastatus disparis*, the larval parasitoids *Cotesia melanoscela*, *Blepharipa pratensis*, and *Parasetigena silvestris*, and the pupal parasitoid *Brachymeria intermedia*. Another introduced larval parasitoid, *Compsilura concinnata*, which has a wide host range, attacks many species of larvae in addition to the gypsy moth. The egg parasite *O. kuvanae* is usually abundant and typically attacks from 10 to 40% of all gypsy moth eggs (Brown 1984). However, because it can reach only the outermost eggs in an egg mass, its effectiveness is limited. The larval parasitoid *C. melanoscela* typically is abundant, but high rates of overwintering mortality and poor synchronization with host development limit its impact.

Simons et al. (1979) provides a guide to gypsy moth parasitoid identification.

Predators

Invertebrate predators: Ground beetles, ants, and spiders are known to feed on gypsy moth larvae and pupae. One predatory beetle, *Calosoma sycophanta*, was successfully introduced into North America from Europe. This ground beetle sometimes becomes abundant in outbreak gypsy moth populations, but usually lags one to three years behind (Weseloh 1985).

Birds: Many species of birds feed on gypsy moths, but they are not a major diet item for any of the common species (Elkinton and Liebhold 1990). Most birds are deterred by the long hairs on larvae. Nuthatches, chickadees, towhees, vireos, orioles, catbirds, robins, and blue jays are probably the most important species in innocuous-phase gypsy moth populations. Cuckoos and flocking species such as starlings, grackles, red-winged blackbirds and crows may be attracted to outbreak populations (Smith and Lautenschlager 1978).

Mammals: Shrews and white-footed mice eat larvae and pupae and may be a major factor in the maintenance of low gypsy moth populations (Elkinton and Liebhold 1990). There is some evidence that regional changes in small mammal density may account for the region-wide onset of gypsy moth outbreaks (Liebhold and Elkinton 1989).

Area-Wide Suppression

***Bacillus thuringiensis*:** This spore-forming bacterium produces a crystalline protein during sporulation that is toxic to the larvae of many species of butterflies and moths, including the gypsy moth. Predators and parasitoids of the gypsy moth are not harmed by the toxin, nor are humans, plants, or other animals. A complete review of the properties and action of *B.t.* toxin can be found in Dubois (1981). *B.t.* is an effective alternative to chemical pesticides when used against the gypsy moth and is currently available in a number of commercial formulations. Label directions should be followed at all times.

Under most conditions, *B.t.* is generally effective at protecting foliage, although it is less effective at reducing populations (Twardus and Machesky 1990). Two applications of *B.t.* separated by three to seven days may increase the effectiveness of the treatment (Webb et al. 1991). Because it is most effective against very young larvae, the first application of *B.t.* should be made when 50% of the larvae are second instars and oak leaves are at least 50% expanded.

More detailed discussions of *B.t.* dose, adjuvants, dilution, and nozzle type and configuration, as well as spray calibration, characterization, and evaluation, can be found in Reardon (1991).

Nucleopolyhedrosis virus (NPV): This virus is the cause of an endemic wilt disease of gypsy moth larvae in the United States and Europe and is a major cause of naturally-occurring gypsy moth population decline. Its effects are most obvious under outbreak conditions, where a high proportion of the larval population may be killed. It is often referred to as "wilt" disease, because of the limp appearance of infected larvae. Infected larvae eventually rupture, releasing a brown fluid containing virus particles. Transmission of the disease occurs within a generation from contact with infected individuals and contaminated surfaces, and to some extent by gypsy moth parasitiods and predators (Podgwaite 1981). Transmission from generation to generation occurs through exposure to contaminated surfaces (Woods et al. 1989).

A review of the natural occurrence, culture, and testing of NPV as an artificially- applied larvacide can be found in Lewis (1981). Gypchek, the NPV product currently registered with the Environmental Protection Agency, is not yet commercially available, although limited quantities are produced by a cooperative APHIS/U.S. Forest Service project (Reardon and Podgwaite 1992). It may be available in 1994. Most of this material is used for testing of new formulations and application technology. How a commercial product would be used is not clear, especially because questions have been raised about its impact on non-target organisms.

Gypsy Moth Pheromone

The chemical structure of the sex pheromone produced by female gypsy moths to attract males, known as disparlure (cis-7,8-epoxy-w-methyloctadecane) was identified in 1970 (Bierl et al. 1970) and can now be synthesized for use in management programs. While disparlure is widely used to monitor adult male population levels (see Population monitoring section), it has also been used to control small populations (e.g., isolated outbreaks along the leading edge of the infestation) by trapping males in pheromone-baited traps and by disrupting mating behavior (Plimmer et al. 1982). Currently, APHIS uses pheromone traps (at a density of three to ten traps per acre) in attempts to eradicate small outbreaks in selected areas of the United States (Anonymous 1990).

Mating disruption for gypsy moth management can be effective in certain situations. It cannot be used in areas which are quarentine regulated or experiencing outbreak population levels. Mating disruption has been used effectively to control new infestations in areas that currently have no gypsy moth problem or on leading edge zones of current infestations. (The 100-mile border of current infestations which border uninfested areas.) Mating disruption is used in areas where there are fewer than 10 egg masses per acre, which corresponds to an average of 20 male moths/trap/season, or a maximum of 40 male moths/trap/season.

There are two types of dispensing systems for the pheromone; a flake formulation, which is currently on the market and a bead formulation, which will be fully registered by the end of 1994. The flake is expensive to apply because specialized aircraft application pods are required. It is long-lasting (eight weeks) and has a steady release rate over that time, so it provides more flexibility in time of application. The bead is less costly to apply, since a regular aircraft spray boom can be used. It is less effective than the flake because it tends to release quickly, so two applications are usually needed. Available in several bead sizes, the smaller bead releases more quickly than the larger. Temperature governs release rate and will be faster in warmer weather and slower under cooler conditions. See Leonard et al. (1989) for more information on this technique.

Genetic Control

The release of sterilized gypsy moths has been attempted as a means of control, but is still in the research and development stage. See Mastro et al. (1981) for a detailed discussion of the USDA sterile gypsy moth release research program.

Favored-Host Removal

Since the demise of the American chestnut as the dominant overstory tree in the eastern United States deciduous forests, oaks have become a dominant species. Unfortunately, oaks are also the favored hosts of the gypsy moth throughout its range. In the absence of external control measures, repeated defoliation of favored trees may result in a shift of dominance to nonhosts and less favored hosts, such as maples. This will ultimately reduce the magnitude of the gypsy moth problem in these areas. While selective removal of favored gypsy moth hosts is an impractical (at best) solution for most park sites, selection of planting material for areas under development (e.g., urban parks) to exclude favored hosts is definitely feasible and should be strongly encouraged.

In managed forests, one option for gypsy moth management that is available to the resource manager is **silvicultural control**. This is the selective harvest of trees to reduce the susceptibility (likelihood of defoliation) and vulnerability (likelihood of mortality after defoliation) of the forest stand to gypsy moth outbreaks. This is done by maximizing tree growth and vigor, removing high-risk trees, manipulating the habitat of the gypsy moth and its natural enemies, and increasing forest diversity. Further discussion and guidelines for silvicultural management can be found in Gottschalk (1993).

Regulatory Control

APHIS has designated most of New England, the mid-Atlantic states, and portions of Michigan as "gypsy moth high risk areas" (Anonymous 1990). Other areas of the United States may be designated by APHIS as high-risk areas if isolated infestations develop there, until gypsy moths are successfully eradicated. Individuals moving household or recreational items from these areas into or through other areas of the United States must have such items inspected and certified ``gypsy-moth-free" by a USDA-trained inspector. Since gypsy moths may be carried on surfaces of vehicles, camping equipment, and other outdoor items, inspection of the vehicles and equipment belonging to park visitors from high-risk areas may enable park personnel to discover and destroy egg masses and other gypsy moth life stages which could give rise to new infestations. Distribution of educational materials (e.g., Don't Move Gypsy Moth [Anonymous 1983]) to prospective visitors of all parks outside high-risk areas, along with the erection of prominent informational displays outside park boundaries, are recommended as methods to encourage visitors to voluntarily participate in such a program. Contact your regional National Park Service Integrated Pest Management coordinator or local APHIS office for help in setting up such a program.

The establishment of a pheromone-trapping program in areas of high vehicular traffic and other visitor use is recommended as an adjunct to any inspection program, to permit the discovery of isolated infestations caused by egg masses or other life stages slipping through the inspection program. Contact your local U. S. Forest Service office for details and assistance in conducting a trapping program.

Currently, APHIS has responsibility for the eradication of isolated infestations of 640 acres or less. Suppression efforts over larger areas and within the generally infested area are the responsibility of the U.S. Forest Service. Some parks receive U.S. Forest Service funds for gypsy moth management and contract for their own management programs. APHIS uses either insecticides, including multiple applications of *B.t.*, or mass trapping, to eradicate isolated populations.

CHEMICAL CONTROL OF GYPSY MOTHS

Several chemical insecticides are currently registered for gypsy moth control. National Park Service policy states that these pesticides may only be used in historic or developed park areas in which *B.t.* or other biological methods (or pheromone trapping) are ineffective. Contact your regional National Park Service Integrated Pest Management coordinator for further information.

Systemic injection: Injections or implants of insecticides registered for this purpose and applied to oak trees at budswell provide significant protection from gypsy moth defoliation (Webb et al. 1988). Some wounding to the tree occurs with this procedure, with white oaks exhibiting a more severe wound response than red oaks (Reardon and Webb 1990). It appears that most of the wounds close and trees recover within three years.

Ground application of insecticides: Individual trees in areas accessible to vehicles can be sprayed with registered insecticides from the ground, using hydraulic sprayers or mist blowers to protect foliage. Although relatively expensive, this method can be quite effective. Since the entire infested area may not be treated, the potential exists for reinfestation of treated trees from the surrounding area. Sticky barrier bands on treated trees may be helpful in preventing reinfestation.

SUMMARY

The U.S. Forest Service is responsible for conducting gypsy moth population monitoring programs on all Federal lands. Each park manager should contact his/her regional U.S. Forest Service office for assistance in setting up an appropriate gypsy moth monitoring program for high-use areas. For further information regarding U.S. Forest Service services, contact:

U.S. Forest Service
Forest Pest Management
1720 Peachtree Road
Atlanta, GA 30367
(404) 374-2989

U.S. Forest Service
Forest Health Protection
5 Radnor Corporate Center
Suite 200
P.O. Box 6775
Radnor, PA 19087
(215) 975-4125

In historic and developed parks (including campgrounds, visitor facilities where shade is an important attraction, and specimen trees), survey programs may trigger suppression or eradication activities. Under National Park Service policy, natural areas, areas containing endangered species, or areas with special natural features may receive **no** treatments; existing natural enemies must be allowed to exert their long- term effects in such areas.

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Leafy Spurge

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for leafy spurge. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Introduction

Leafy spurge (*Euphorbia esula* L.), a member of the family Euphorbiaceae, is a herbaceous, deep-rooted perennial weed of disturbed lands. Pastures and fields left fallow for long periods, tree rows, waste areas, roadsides, and rangelands or open grasslands are all susceptible to infestation by leafy spurge. Leafy spurge commonly occurs along railroad rights-of-way, water courses, and gullies. It is sometimes found in cultivated lands where infested land has been broken for crop production. Leafy spurge rarely invades fields that have been under cultivation for several years, but long-lived roots can regenerate at any time. The single greatest direct impact of leafy spurge is the reduction of populations of native grasses and legumes and associated ecosystem changes caused by the superior competitive abilities (rapid growth and allelopathic properties) of this species (Steenhagen and Zimdahl 1979).

Indirect impacts of leafy spurge infestations include the loss of food sources for grazing animals caused by competition with native plants in pastures and on rangeland. Leafy spurge infestations may cut pasture production by 50%-75%. Since wildlife and cattle generally avoid grazing in infested areas, carrying capacity may be reduced by up to 75% (Lacey et al. 1984). Leafy spurge is toxic to most grazing mammals, and the milky latex contains substances that act as irritants, emetics, and purgatives for many animals when eaten.

A second indirect impact of leafy spurge is the cost of control; in some cases, the cost of control may exceed the original cost of the land (Lavigne 1984). Due to the extremely deep and hardy root system, control of established leafy spurge populations in uncultivated areas is costly and control measures must take place continuously over several years. Leafy spurge often regenerates when controls are eased. Because of its economic importance, leafy spurge is listed as a noxious weed in many states, with control legally mandated.

Although the competitiveness and toxicity of leafy spurge makes it undesirable, its pest status, as with most introduced species, results from the lack of population suppression exerted by natural enemies (e.g., insect herbivores and diseases). Thus, long term goals in leafy spurge management emphasize biological and cultural controls, although emergency intervention with chemical or mechanical controls may be necessary. Leafy spurge is fairly easy to control within the first two years of establishment. After the third or fourth year, the root system becomes so well developed that the plants are little affected by mowing, cultivation, grazing, or pulling, and herbicides cannot be translocated to the deepest portions of the roots.

IDENTIFICATION AND BIOLOGY OF LEAFY SPURGE

Leafy spurge was introduced into eastern North America from the Old World in the early 19th century. Other introductions were made in the midwestern United States in the late 19th century, probably as contaminants in seed grain (Messersmith and Lym 1983a). Many reintroductions and crosses have occurred in other areas and at other times, resulting in a highly diverse and complex population throughout North America (Lorenz and Messersmith 1981). The extreme plasticity and genetic diversity of this plant has led several researchers to believe that the leafy spurge of North America is a hybrid between two or more Old World spurges (Schaeffer and Gerhardt 1984).

Leafy spurge reproduces by seed and from spreading roots. The roots are deep, woody, and very persistent. Stems are erect, glabrous, branched at the top, and contain a milky sap. Leaves are alternate, broadly linear to narrowly oblong-lanceolate. The inflorescence is a terminal open umbel of greenish flowers, each about 1/8 " high. The petals are fused into a cuplike structure, borne just above the greenish-yellow heart-shaped floral bracts on the top of the stem. See Messersmith (1983) and Eberlein et al. (1982) for complete descriptions and photographs of leafy spurge.

Germination from overwintered seed occurs in early May. True leaves appear 6-10 days after germination. The first pair of true leaves are opposite; later, all leaves are alternate. Stem elongation and vegetative growth occur in mid-May.

Leafy spurge produces vegetative stems from existing roots in late April, making leafy spurge one of the first plants to emerge in spring. Early and rapid growth gives leafy spurge a competitive advantage over most crop and pasture plants.

Yellow bracts form in late May, with maximum display from early to mid- June. Flower development is through mid-June, and the first fully developed seeds occur in early July. Seeds are borne in groups of three within each pod. Seed dispersal is in mid-July, during hot, dry weather. Pods burst violently, scattering seeds up to 15' away from the parent plant. The seeds float and are frequently dispersed by streams.

Leaf loss and late summer dormancy occur during late July to mid-September. Plants renew growth in mid-September with the advent of cooler weather. Several leafy branches are formed off the main stem, which remains leafless. During this period, photosynthesis resumes and additional photosynthates are transported to the root system for storage through the spring.

The root system is extensive, and consists of numerous coarse and fine roots which occupy a large volume of soil. Roots are most abundant in the upper foot of soil, but some roots can extend to a depth of 30'. The root system contains a large nutrient reserve capable of sustaining the plant for years. Root fragments as small as 1/2" long can give rise to new plants. Leafy spurge can withstand repeated mowing and cultivation (Eberlein et al. 1982) due to this well-developed food storage system in the roots. Roots have the ability to regenerate plants from almost any depth.

Leafy spurge usually forms patches that may reach a density of over 200 stems per square yard in sandy soils and higher in heavy clay soils. Patches of leafy spurge usually spread vegetatively at a rate of 1' to 3' per year with allelopathic chemicals secreted by the root to reduce competition (Eberein et al. 1982). Plants emerge in April (from root stocks) or May (from seed) and persist throughout the growing season.

MONITORING AND THRESHOLDS FOR LEAFY SPURGE

Leafy spurge populations are best monitored when the plant is most conspicuous, i.e., when the yellow green flower-like bracts are open in late May to mid-June. Because leafy spurge usually occurs in patches, monitoring usually involves counting or estimating the number of patches per unit area (acre, hectare, etc.). Calculate the average patch size, and count the number of plants per square yard or meter in sample patches. This will give a fairly accurate estimate of the number of plants per area. Careful records should be kept in order to establish a profile of infestation patterns, rates, and treatments.

Leafy spurge can be monitored by aerial infrared imagery using Kodak 1443 color infrared film (for mapping purposes, use large format 9x9 2443 film), a yellow #12 filter, and a film scale of 1:24,000 or larger. Leafy spurge should be in full "bloom" (bract display) and growing vigorously during the second week of June to the second week of July. The image on false color infrared film will be hot pink, which is characteristic of leafy spurge at full bloom and not easily confused with any other plant. Patches as small as 10' x 10' (100 ft^2) are easily identified using this method. See Armstrong (1979) for further details.

Economic thresholds for leafy spurge have not been developed. While it is known that heavy infestations can lower range productivity, the cost of mechanical and chemical controls are often considered to be uneconomical in most of the affected areas (Sun 1981). Most ranchers consider spurge to be below injury level if spurge patches do not expand from year to year. In natural areas within a park, leafy spurge management should begin when an infestation is discovered. In areas such as historic or developed sites, or where park lands are adjacent to private or public grazing lands, management techniques should be employed to prevent spurge infestations, and established patches should be controlled to prevent spreading.

NON-CHEMICAL CONTROL OF LEAFY SPURGE

Leafy spurge is difficult to eradicate, but control is possible if a persistent management program is followed. Control strategies should focus on containing the spread of populations by treatment of new populations within their first two years of establishment, and also on concentrating efforts on the advancing edges of established spurge populations. Treatments of well-established plants should receive lower priority. Long-term strategies for weed control depend on biological and cultural controls, while chemical and mechanical controls are useful for short-term suppression. Generally, no one technique will provide adequate control. Currently available biological controls using insects require several years for establishment of the insect, and even longer for control. Most successful programs combine biological control with cultural controls such as timely mowing or reseeding with competitive desirable plants. Suppression of leafy spurge may require altering land use.

Biological Control

Herbivorous insects. Leafy spurge is attacked in North America by only a few generalist native herbivorous insects (Harris 1979). Consequently, natural enemies of *E. esula* in Europe and Asia have been imported to the United States and Canada. However, hybridization and other factors are believed to have changed the genotype of the North American spurge, and as a result, most natural enemies from its area of origin have had inconclusive results in North America.

The spurge hawk moth, *Hyles euphorbiae* (L), (family Sphingidae), was introduced into Canada in 1977 (Forwood and McCarty 1980). Populations stabilized at densities considered too low to provide effective control, however, and eventually declined to extinction. Subsequent introductions in Montana and New York have become established and introductions may occur in other states. The spurge hawk moth has one generation per year. Although caterpillars defoliate plants, leafy spurge foliage usually regenerates.

Negative results have been obtained with most introductions. The moth *Chamaesphecia tenthrediniformis* (Denis & Schoff) was released in Canada in 1970 after promising results in feeding tests. However, all larvae released in the field died without feeding on leafy spurge. The Canadian release of the aphid *Acrythosiphum neerlandicum*, which is only known from *E. esula* in Europe, resulted in death on Canadian leafy spurge (Harris 1979).

The stem-and root-boring cerambycid beetle, *Oberea erythrocephala* (Schrank.), which attacks both *E. esula* and *E. virgata* , has been released and established in the western United States (Rees et al. 1986). The main influence on leafy spurge is a reduction in number and vigor of stems produced in the following year. Long-term effects are not known.

The flea beetle, *Aphthona flava* Guill., feeds as an adult on the leaves of leafy spurge, causing minor damage. The larvae feed heavily on the roots, causing stunting and eventually killing the plant. There is one generation per year. This species has been established in the U.S. and Canada (McClay and Harris 1984, Pemberton and Rees 1990).

The cecidomyid gall midge, *Spurgia esulae* Gagne, which forms galls over the branch tips that slow growth, stunt the plant, and prevent blossoming, has been evaluated and released (Pecora et al. 1991). This species has several generations per year, making it an excellent potential biological control agent.

Grazing by sheep. Although grazing by livestock has not been recommended in the past, Landgraf et al. (1984) have found that sheep may graze on leafy spurge without ill effects. The diet of sheep can contain up to 50% leafy spurge with no significant difference in weight gain compared to sheep feeding in spurge-free pastures. They conclude that sheep are a viable biological control agent for leafy spurge. Pastures grazed by sheep from May to September for five successive seasons show up to 98% reduction in spurge populations. Utilization of and effects of leafy spurge on lambs and lactating ewes has not yet been quantified. Grazing by sheep may not be an appropriate control measure in natural areas. Some varieties of spurge may be rejected by sheep, and in most cases spurge will regenerate the season after grazing pressure ceases.

If sheep are to be used as a biological control for leafy spurge, the following guidelines from Lacey et al. (1984) should be followed:

- Grazing should begin in the spring when spurge plants are only a few inches tall.
- Schedule sheep grazing rotations so that spurge does not go to seed.
- If sheep graze after seed set, animals should be held for five days to allow viable seeds to be passed before sheep are moved to new pastures.
- Sheep grazing can be combined with herbicide use around the fringes of patches for optimal control.

Pathogens. Several plant pathogens have been tested on leafy spurge, including rust fungi, powdery mildews, soil borne fungi, and foliar pathogens. To date, none have been found to be desirable control agents due to wide host ranges (which include domestic crops) or lack of permanent control. Several rusts and *Alternaria* species have been collected recently in Europe and are undergoing testing at this time (Littlefield 1984, Yang et al. 1990).

Cultural Control

In areas where planting of competitive crops is possible, crops such as sudangrass or buckwheat may be utilized. Competitive cropping reduced leafy spurge stands by 50% in the first year of trials, and 80% in the second year when given three cultivations before seeding, and with stubble plowed after harvest (Derscheid 1979).

Elimination of leafy spurge was also achieved in two years following planting of close-drilled forage sorghum or soybeans. A short season of intensive cultivation followed by planting of fall seeded crops of bromegrass reduced leafy spurge populations by 95% (Derscheid 1979). Crested wheatgrass also competes successfully with spurge, but it should be noted that bromegrass and crested wheatgrass are exotic species that are generally considered inappropriate for natural areas. Reinfestation of leafy spurge from seed can be prevented by using soil-building crop rotations. Legumes (such as sweetclover) will prevent establishment by most leafy spurge seedlings (Derscheid 1979).

Mechanical Control

Use of controlled burning has been attempted in North Dakota and in Wyoming. Although burning has little effect on established plants with deep root systems, fire may be highly effective in reducing seed and seedling viability. Controlled burns in the fall against the wind (burning against the wind results in more complete combustion and hotter fires) resulted in reduced germination rates.

Mowing, especially when used prior to treatments with herbicides, may allow reduced rates of chemicals to provide effective shoot control (Ferrell and Alley 1984b). Hand pulling of leafy spurge while in the bloom stage results in reduced regrowth vigor for two years. Pulling also damages the root, increasing the chance of infection by pathogenic organisms (Maxwell et al. 1984).

Intensive cultivation at 2-3 week intervals will reduce leafy spurge stands by 90% in the first year, and give complete control in 2 years. Similar results have been achieved by cultivation with a duckfoot cultivator every 2-3 weeks or a springtooth harrow each week (Derscheid 1979).

CHEMICAL CONTROL OF LEAFY SPURGE

The use of herbicides provides a quick and easy (albeit expensive on large- scale operations) method of control. Herbicides, applied prior to flowering, give excellent burn-down of top growth but no long-term control of well-established plants. Without a long-term strategy, herbicides often lead to greater problems in the future because of their effect on other plant species, the development of resistance, and the inability to completely eradicate populations.

Consult your regional Integrated Pest Management coordinator to determine which, if any, herbicide is best suited to your integrated pest management program.

SUMMARY

To summarize, the following steps are recommended to manage leafy spurge:

1. Monitor leafy spurge by ground checks or aerial surveys using false color infrared film.

2. Determine injury levels based on land usage (local weed ordinances should be acknowledged).

3. Control strategies should focus on containing the spread of populations by treatment of new populations within their first two years of establishment and also to concentrate efforts on the advancing edges of established spurge populations. Treatments of well- established plants should receive lower priority.

4. Use cultural or mechanical controls to reduce small to medium infestations. Consider the use of controlled grazing by sheep as a biological control.

5. Use registered herbicides where appropriate; applications should be timed for best control, and follow-up treatments should be applied when necessary.

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Mites

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for mites. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Mites are members of the order Acarina, which also includes ticks. Hundreds of species of mites occur in the United States. This module describes life histories and integrated pest management strategies for seven species that have been found to be of greatest concern in the National Park System. Six of the mite species in this package are in the family Tetranychidae (which includes the mites commonly known as spider mites), while the seventh, the eriophyid mites, are in the family Eriophyidae. All are extremely small, requiring a hand lens to determine their presence and numbers. Mites do not have a true head, wings, or abdomen. There are four pairs of legs and a pair of leg-like palps associated with the mouthparts. Mouthparts consist of a pair of needle-like stylets (chelicerae) used to pierce cell walls, allowing the mouth to suck up cell contents. This is important because the type of mouthpart creates the stippled appearance associated with Tetranychid mite injury. The injury caused by Eriophyid mites is much more variable, and includes yellowed foliage, distorted foliage, or a variety of leaf and petiole galls. Mite-feeding injury is often confused with injury caused by insects or air pollution. Refer to Table 1 for more information on differentiating mite and insect injury.

Table 1. Plant pests and diseases which produce injury similar to Mites

Pest	Symptoms	Detection	Control/Prevention
Teranychid mites	Leaves or needles get pale yellow to bronze stippled areas at any time from early spring on. Usually on one-year-old growth of conifers and new growth of broad-leaved and deciduous plants.	Tap branches onto white paper and examine with a 10x hand lens for small green or red spiders or shiny round eggs. White cast skins may be seen on leaf undersides. Look for eggs at any time, mites when temperatures are above 50 degrees.	Dormant oil in late winter; insecticidal soap, summer oil or miticide at other times as needed. Ref: Grossman; Johnson and Lyon
Eriophyid mites	Yellow to bronze areas at tips of needles or twisted, distorted foliage, or witches broom. On hemlock, entire plant becomes chlorotic. Many types of leaf or petiole galls on deciduous plants.	Look for clear, cigar-shaped mites with a 10x hand lens or microscope. On conifers, they are at the base of the needle or under the needle sheath, except on hemlock, where they are found on all parts of needle underside. They are	Insecticide or miticide when present. Ref: Johnson and Lyone; Kelfer and Smith

		difficult to detect	
Lace bugs	Leaves have yellow to bronze speckled appearance with tar spots on leaf undersides.	Look for symptoms throughout season.	Don’t plant host plants in full sun; apply insecticidal soap or registered insecticide when nymphs or adults are active. Ref: Johnston and Lyon
Leafhoppers/Planthoppers	Stippled, bleached, or distorted foliage	Observation of the pest of its cast skin in mid- and late summer	Registered insecticide when insects are active. Ref: Johnson and Lyon
Thrips	Stippled or bleached upper foliage or flowers, often with black frass on injured areas. Stippling often seen in rows.	Stippling appears from spring on, depending on region and thirps species. Examination of stippled areas with a 10x hand lens shows that leaf tissue has been scraped away.	Registered insecticide when insects are active. Ref: Johnson and Lyon.
Ozone	Large bronze to brown stippled areas seen only on upper surface of leaf.	Visual observation of injury is related to a period of poor air quality or a termperature inversion	Replace with a resistant variety or species if one is available. Ref: Sinclair et al

IDENTIFICATION AND BIOLOGY OF TETRANYCHID MITES

The life cycle of tetranychid mites includes the following stages: egg, larvae, nymph (up to several nymphal instars), and adults. In some species, males are unknown and reproduction is believed to be parthenogenic (Johnson and Lyon 1988; Weidhaas 1979). This means that females give rise to offspring without mating, enabling rapid reproduction and population increases. Because of the large number of generations in a single season, high infestations of mites can develop rapidly (Huffaker et al. 1969; Johnson and Lyon 1988; Helle and Sabelis 1985; Westcott 1973; Yepsen 1984). In general, mites deposit two to twenty eggs in a single day, the exact number determined by environmental factors and the species or strain involved. Silk production by mites varies from species to species, with some producing copious amounts of silk, others little or none.

Habitats for the mite species in this module consist mainly of the foliage of suitable host plants. Larvae and nymphs tend to feed on the underside of leaves, while adults and older nymphs feed on both undersides and tops of leaves as well as occasionally on buds and shoots.

Mites feed by rupturing leaf cells with a pair of needle-like stylets (chelicerae) and inserting the mouth parts to draw up the cell contents while the chelicerae are pushed deeper. Feeding causes small chlorotic spots to appear, which eventually coalesce. Stippling occurs and large portions of the leaf or the entire leaf becomes yellowed, bronzed, or whitened in appearance. Leaf injury on evergreens may last for several seasons; injury on other plants may cause premature leaf drop or may result in the death of the host plant.

Boxwood mite (*Eurytetranychus buxi* [Garman]). Adults are 1/32" long, yellow green to reddish brown. Eggs are yellow, rounded, with flattened ends. This species produces silk and is found throughout the United States on boxwood, specifically varieties of American and European boxwood (*Buxus sempervirens*). Japanese boxwood (*B. microphylla*) is rarely infested. The boxwood mite eggs overwinter on the undersides of leaves and hatch in mid-April. Early nymphs feed on undersides of leaves, second instar mites feed on both sides of the leaf, and third instars move from leaf to leaf to feed. Adults feed on shoots and upper surfaces of leaves. Populations are highest from early spring to early summer, with a second peak in the fall.

Clover mite (*Bryobia praetiosa* [Koch]). Adults are brownish red to red, 1/16" in length. Eggs are brick red, the nymphs red. These mites are easily recognized under low magnification by their long front legs, which are over twice as long as the other legs, and by the featherlike plates on the body. This species does not produce silk, so the presence of webbing cannot be used as a sign of this pest. Distributed throughout the United States on suitable host plants, clover mites are also common indoors, frequently entering buildings in large numbers in the fall.

Clover mite eggs overwinter in cracks in concrete foundations, between the exterior and interior walls of buildings, and on the underside of the basal bark of trees. These mites also overwinter as adult females or in other life stages. Clover mites can become active at temperatures slightly above freezing. Eggs hatch in late winter or early spring; one generation is usually complete before mid-summer. Males of this species are unknown; reproduction is parthenogenetic (Boudreaux 1963). Most eggs deposited by this generation aestivate until September, but some hatch in early summer and produce several small successive summer generations.

These mites feed on a wide variety of plants including clover, grasses, dandelion, iris, ivy, mallow, strawberry, peas, tomato, violet, and zinnia. A related species, the brown mite, feeds on tree foliage.

European red mite (*Panonychus ulmi* [Koch]). Adult mites are 1/32" long and velvety red, with four rows of curved hairs on back arising from tan or white humps (tubercles). Eggs and first instar nymphs are bright red; each egg has a single central stalk or hair. Second and third instar nymphs are dull green or brown. This species produces silk and thus webbing is seen at high population levels. European red mites occur throughout the United States on suitable host plants. They feed on apples and other fruits, nuts, and their ornamental varieties. They may occasionally be found on elm, rose, mountain ash, and a variety of other ornamental plants. European red mite overwinter as eggs and hatch in early spring as new growth begins. Feeding activity and plant injury occur throughout spring into early summer.

Southern red mite (*Oligonychus illcis* [McGregor]). Adult females are 1/32" in length, blackish red, with backward curving spines. Adult males, nymphs, and eggs are light red. This species produces silk. They are common in the southeastern United States, New England, Ohio, and the Great Lakes states but are particularly damaging in the deep south. Southern red mite feed on broad-leaved evergreens, especially Japanese holly, *Pyracantha*, azalea, and *Camellia*, as well as other hollies, laurel, and *Rhododendron*. They overwinter as eggs on the foliage and twigs of their hosts. A cool weather pest, Southern red mite develop damaging populations in early spring and late fall. These mites thought to aestivate in the egg stage during summer, with small populations becoming active during cool periods.

Spruce mite (*Oligonychus ununguis* [Jacobi]). Adults are 1/32" in length with spines on the back, dark green or reddish green to nearly black with tan legs. Eggs are reddish tan and nymphs greenish with tan legs. Spruce mites produce copious webbing between needles of host plants. They are widely distributed and may be found wherever suitable hosts occur. They attack only conifers; primarily hemlock, spruce, arborvitae, *Chamaecyparis,* and juniper. Fir and pine are attacked to a lesser extent. This mite overwinters as eggs on the foliage and twigs of host plants. They are most active in cool weather, so tend to increase in numbers and injury levels in early spring to early summer, and again in the fall, while they may go into aestivation to avoid hot, dry weather. Adults may be active in summer during cooler periods.

Twospotted mite (*Tetranychus urticae* ([Koch]). The common "spider mite." Adults are large (1/8"), and yellowish with two or more predominant dark spots on the back, which is sparsely covered with spines. These spots, which become more apparent as each instar matures, are caused by accumulated food material in the digestive tract. The eggs and nymphs are lemon yellow. They are found throughout the United States, especially indoors and in greenhouses. There are over 250 known host plants including flowers, foliage plants, corn and other field crops, vegetables, brambles, and other herbaceous plants. They can be a serious pest of roses, flowering fruits, and shrubs, and are frequently brought outdoors on plants which were propagated or overwintered in the greenhouse. They overwinter as eggs on host plants and cause damage to host plants throughout the growing season. The warmer the temperature, the greater the rate of feeding and reproduction. The twospotted mite becomes especially destructive during periods of hot, dry weather, but also feeds and reproduces during cooler periods.

The twospotted mite can acquire several plant-infecting viruses during feeding on infected hosts, but has not been shown to transmit them to new host plants (Orlab 1968). Mites that enter houses can create a nuisance to homeowners and can cause stains if they are crushed.

MONITORING AND THRESHOLDS FOR TETRANYCHID MITES

Mite population cycles can be unpredictable, so timing of management practices must be based on observations of the pest. Timing of monitoring for mites is directly related to mite biology. For example, spruce mites may be active anytime temperatures are over 50F, but once prolonged, hot, dry weather occurs in summer they enter a type of dormancy known as aestivation and generally do not become active again until fall. Aestivation occurs at about the end of June in the mid-Atlantic region, when daytime temperatures are consistently above 80F. Spruce mite aestivation corresponds to the time when the activity level and generation times of other mite species, such as the twospotted mite, the European red mite, and the southern red mite, are increasing. Consult the information presented for each species as well as the references for more detail on the population cycles of each mite species.

Mites are very small, so they must be knocked off the plants they are feeding on to be counted. This is done by holding a piece of white paper or a clipboard painted white under the plant and striking the branches with a rubber hose or ruler. Generally the plant is struck three to five times before the mites are counted. The number of times that this is done is not as important as doing it the same number every time. Ten to fifteen seconds must pass before examining the clipboard for mites, since it takes this long for them to begin moving after being knocked off their host. Moving dots about the size of a period on this page should be examined with a hand lens to determine that they are indeed mites and to identify the species if possible. Population levels can be measured in a variety of ways, including a simple presence/absence, ranges (e.g., 1-10, 11-20, etc.), or actual population counts. In most cases, estimation of a population range will suffice. Eggs tend to remain attached to the plant, so individual branches must be examined to look for these. Again, estimating relative numbers is more important than an absolute count. The number of eggs relative to the number of adults will indicate if the population is increasing or decreasing.

Mite populations on plants that have cupped leaves, such as Japanese holly, also need to be determined by examination of the individual plant, since mites tend to remain in the cupped leaves when the plant is tapped.

Monitoring for mites is a time-consuming process. If you are willing to tolerate some mite injury, the time required for monitoring can be reduced by focusing monitoring efforts on plants in hot, dry areas, plants which have been under heavy nitrogen fertilization, or plants which have had the most serious problems in the past. If low mite populations are seen on these plants, then monitoring of less susceptible plants can be skipped **at that time**. This is not recommended if the aesthetic threshold of the plants being monitored is very low (i.e. no injury can be tolerated).

Leaf discoloration and stippling caused by mite feeding can easily be confused with several other insect and disease injury symptoms (refer to Table 1). It is important not to assume that just because stippling is seen, mites are the cause. Mites, eggs, or shed skins on leaf undersides will facilitate a diagnosis. Stippling with tar-like frass on leaf undersides indicates lace bug; lack of frass or mite signs is a clue that the injury is from air pollution.

Once mite activity is detected, a decision must be made as to whether implementation of additional mite management tactics are warranted, and if so, which is most appropriate. While a few action levels for mites have been published (Hamlen et al. 1982; Nielsen 1989), it is unclear how these apply in a generalized way. There is considerable evidence to show that host plant nutrient status and drought stress (Jepsen et al. 1975; Mattson 1980) contribute to host plant suitability as a food source for mites. This means that action thresholds determined under one set of conditions may not be applicable in another system. Published thresholds should be used as a guide, but to modified as the need arises. The resource manager must keep accurate records of mite population levels, plant injury symptoms, soil fertility, and rainfall at the individual park. Relate these to timing and type of management strategies used in the past to determine what works best at a particular site or on a certain plant species.

NON-CHEMICAL CONTROL OF TETRANYCHID MITES

Good mite management combines regular monitoring to detect pest occurrence and **timely** implementation of the most appropriate management tactics. Monitoring is an essential part of a mite integrated pest management program because injury cannot be seen until after feeding takes place and because mites may be active any time microhabitat temperatures rise above 50F. This means that even though the ambient air temperatures are below 50F, mites could be active on certain plants, such as those in sunny locations next to a building.

Cultural Control

As was mentioned earlier, there is a considerable amount of evidence to indicate that mite populations are higher on plants that have been under high nitrogen fertilization regimes (Mattson 1980). Thus, plants that have mites should not be heavily fertilized.

Physical Control

A strong, steady stream of water from a hose will wash mites from the surface of some plant leaves. Prolonged (several hours) periods of heavy rain have the same effect. This is only a temporary measure, most suited to an area where no pesticides can be applied. Adult mites will generally return to the plant within 24 hours.

Biological Control

A vast number of predators and pathogens have been examined for their potential to serve as biological control agents for mites (Helle and Sabelis 1985). Some are currently being successfully used, others show potential, while the feasibility of others seems unlikely.

Mites in the family Phytoseiidae are important predators of plant-feeding mites and have been used in biological programs for several pest species, particularly in greenhouses. Spiders, beetles, flies, thrips, true bugs, and lacewings have all been observed feeding on mites. Species in the lady beetle genus *Stethorus* are voracious predators of mites and often eliminate infestations of European red mite and spruce mite. However, the control often occurs after the mite populations have peaked (Johnson and Lyon 1988). Tetranychid mites are also susceptible to fungal and virus infections, but no pathogenic bacteria have been reported as occurring in mites. There are no known insect parasitoids of mites (Helle and Sabelis 1985).

Predaceous mites have been used in greenhouses to control twospotted and other mite pests with good results. Predatory mites are available from commercial suppliers. See Anonymous 1991 for a list of sources. Some commonly used predatory mites include the following.

Phytoseiulus persimilis is a predatory mite used primarily in Europe to control mite pests of greenhouse-grown tomatoes, cucumbers, and sweet peppers. It must be released periodically at carefully timed intervals for optimal control. It is used infrequently on greenhouse-grown ornamentals due to lower damage tolerance levels and lack of resistance to pesticides (Field and Hoy 1984).

Insecticidal soap is toxic to the adult predatory mite at rates needed to obtain satisfactory phytophagous mite control. Insecticidal soap can be applied three days after predator release without significantly reducing control, apparently because it does not cause significant egg mortality (Osborne and Petitt 1985). A recent study of the effect of abamectin (a pesticide derived from a bacterial toxin) on this mite and the pest mite, *Tetranychus urticae*, demonstrated that abamectin will reduce the population of both, with a greater reduction in the population of the pest mite species than in the predatory mite. Thus it could be used in an integrated pest management program to reduce the predator/prey ratio and increase the effectiveness of *Phytoseiulus persimilis* as a predator (Zhang and Sanderson 1990).

Phytoseiulus macropilis, a related predatory mite, was found by Hamlen and Poole (1982) to give acceptable control on twospotted mite on greenhouse-grown Diffenbachia when applied at a ratio of 1:10 or lower and reintroduced every eight weeks. As with *P. persimilis*, predators must be introduced into low-density spider mite populations (Samlen and Lindquist 1981).

Mataseiulus* (*Typholdromus*) *occidentalis is a predatory mite that has been developed into several strains, one of which is resistant to most organophosphate insecticides and to carbaryl. Another strain does not go into dormancy under low light or short photoperiod conditions. These strains are preferred in that they can prey upon twospotted-mites throughout the year in greenhouses. *M. occidentalis* is preferred for mite control for ornamentals and long-term crops (such as roses grown for cut flowers) because it gives long-term control from a single release. This predator is unlikely to bring about full control without leaf damage caused by the pest mite; therefore, application of selective acaricides are useful in an integrated program (Field and Hoy 1984). Field and Hoy (1986) suggest that although this species does not give as good control of twospotted mite as does *P. persimilis*, it would be a better choice as a biological control agent in long-term crops on which pesticides will be used. They suggest that *P. persimilis* would be a better predator for twospotted mite on short-term crops that are grown with minimal pesticide inputs. For current information on pesticide resistance in *M. occidentalis*, see Hoy and Conley (1987).

CHEMICAL CONTROL OF TETRANYCHID MITES

Recent advances in the development of horticultural oils have made this the first pesticide to consider in the management of mites (Baxendale and Johnson 1988; Baxendale and Johnson 1989; Davidson 1990; Davidson et al. 1990; Grossman 1990). New oil formulations do not have the problems of phytotoxicity that were so common among older formulations. Their effective control of mite populations with minimal impact on beneficials make them well-suited to an integrated pest management program. A drawback to the use of oils is the necessity of contacting the pest to be killed. Thus oils tend to give unsatisfactory control in dense plantings, on leaves that are cupped (e.g., *Ilex crenata* `Convexa'), or on plants that are in hard-to-reach areas. In these instances, pesticides with some residual activity could be used. Consult your regional Integrated Pest Management coordinator concerning the best choice of pesticide for your situation.

IDENTIFICATION AND BIOLOGY OF ERIOPHYID MITES

Eriophyid mites are a diverse family of arthropods, containing many species with a wide range of plant hosts and biologies. They can be divided into three categories based on the type of plant injury they cause: galls, twisted, distorted foliage and chlorotic, stunted growth. The gall makers are rarely detrimental to plant health, but are a concern among the public because they are so obvious. In general, the mites that cause these galls overwinter as adults and begin feeding on expanding leaves in the spring. This induces formation of a gall which surrounds the mite as it feeds. Eggs are laid within the gall; nymphs mature within the gall and the emerging adults infest new foliage.

The eriophyid mites that injure foliage have varied life cycles. They are a more serious concern than the gall-makers because they can cause distortion and dieback of plant tissue. Consult Johnson and Lyon (1988) and Keifer et al. (1982) for more information on eriophyid mite life cycles.

MONITORING AND THRESHOLDS FOR ERIOPHYID MITES

Eriophyid mites are often overlooked because they are so difficult to see and because the injury they cause (especially necrosis and dieback) can be attributed to many other causes. Thus, the first part of developing a management strategy for eriophyid mites is the education of plant monitors about this mite's biology and preferred hosts and the injury it causes. Monitors should realize that they will most likely not see eriophyid mites without a microscope, and that they may need to submit a sample to the Cooperative Extension Service for identification.

It is difficult to outline a monitoring program for these mites because the life cycles vary so much depending on the species. In general, monitors should be aware of the types of injury caused by eriophyid mites, and that the mites will be difficult to observe. In conifers, this is complicated by the fact that these mites often feed below the needle sheaths.

CONTROL OF ERIOPHYID MITES

In the case of the gall-making eriophyid mites, no intervention is warranted. Three cases where intervention often is appropriate is on hemlock, privet, and white pine, where these mites can cause considerable injury. In these cases, an insecticide such as acephate is usually recommended (Davidson et al. 1990; Keifer et al. 1982; Smith 1990), since they seem to give better control than oil or other miticides.

There have been many observations of predatory mites occurring in conjunction with eriophyids, but their role in population regulation is unknown (Johnson and Lyon 1988).

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Mosquitoes

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for mosquitoes. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

While mosquitoes remain a major killer in other parts of the world, in the United States, mosquitoes are not the scourge they once were. But they're still irritating, they still bite us, and there are some species in the United States that spread disease. Mosquitoes also serve a vital ecological function. The larvae, pupae, and adults are important as food for fish, birds, bats, frogs, and insects- -an essential consideration when the subject of mosquito control arises in a national park.

The one thing that all mosquitoes require to complete their life cycle is water. If people could manage all standing water, we could manage mosquitoes. While we can fill in a puddle, we don't want to fill in a salt marsh. We can empty a bucket, but it's not so easy to empty a tire dump.

BIOLOGY AND IDENTIFICATION OF MOSQUITOES

Pest Species of Mosquitoes

While there are more than 13 genera of mosquitoes in the United States, most pest mosquitoes belong to one of three: *Aedes*, *Culex* , or *Anopheles*. See Figure 1 for drawings of the life stages of these three species.

Aedes

Some *Aedes* mosquitoes are called "floodwater mosquitoes" because they lay their eggs singly on damp soil or vegetation in areas that are periodically wet. The eggs can remain dormant until they are flooded and conditions are favorable for hatching. Salt marsh species breed in coastal marshes that are occasionally flooded by high tides. Many floodwater and salt marsh species can fly great distances (5 to 20 miles) from their hatch site.

Other *Aedes* species prefer to lay their eggs in artificial containers or tree holes. Again the eggs are laid just above the water line and hatch once they are inundated.

The Asian tiger mosquito, *Aedes albopictus*, first appeared in the United States in 1985. Its rapid spread is of concern because it is known as a disease-carrying mosquito in its native Asia. It also breeds readily in water-filled containers, so breeding sites are commonly available.

Culex

These mosquitoes breed in quiet standing water of all types, ranging from containers to larger pools. *Culex* species prefer polluted standing water with a large amount of organic material. Eggs are laid on the surface of the water in "rafts," usually of 100 or more eggs. While *Aedes* and *Anopheles* mosquitoes have a pointed tip at the end of the abdomen, *Culex* mosquitoes have a blunt tip.

Anopheles

Anopheles mosquitoes breed in permanent bodies of fresh water. They prefer water with abundant aquatic plants that provide protection from fish and other predators. Eggs, supported by floats on each side, are laid singly on the surface of the water.

Anopheles mosquitoes can be distinguished from *Aedes* and *Culex* mosquitoes in several ways: (1) Anopheles have patterned wings, (2) adult *Anopheles* females have palps that are almost as long as their proboscis, (3) adults rest on surfaces with their head lower than the abdomen while *Aedes* and *Culex* species rest with the head and abdomen parallel to the surface, and (4) the *Anopheles* larvae float parallel to the water surface rather than hanging down at an angle.

Life Cycle

Of the four life stages of the mosquito--egg, larva, pupa, and adult--the adult is the only stage that doesn't exist in standing water.

The female mosquito lays her eggs on the water or, in the case of *Aedes* mosquitoes, above the water in areas that are sheltered from waves and with sufficient organic matter to feed the larvae. Eggs laid on the water's surface hatch in one to three days. Eggs laid by *Aedes* mosquitoes above the water line remain dormant until they are flooded.

The larvae or "wigglers" that hatch must live in water to survive. They float at the surface breathing through an air tube and filtering food material through their mouth brushes. When disturbed, they dive towards the bottom with a jerking motion. The larval stage lasts from five days to several weeks depending on the species and on environmental conditions such as water temperature.

The larvae transform into pupae or "tumblers." Although the pupae don't feed, they are quite active and may be seen breathing at the surface or bobbing through the water. Inside the pupal skin, the adult mosquito is developing and will emerge in two to three days. Mosquitoes pass the winter either in the egg stage or as adults.

Feeding Habits

Only the female mosquito sucks blood, which she needs to lay eggs. Adult male mosquitoes feed only on plant nectar and are harmless to people.

Most mosquitoes feed just after dark and again just before daylight. They spend the daylight hours resting in dark, damp areas. Some mosquito species, however, feed during the day and others may feed during both day and night.

This blood-sucking habit is what causes certain species of mosquitoes to be disease vectors. If a female mosquito sucks blood from a person infected with malaria, for instance, the disease organisms survive and reproduce in the mosquito, ending up in her salivary glands. When she next feeds on a host, she inoculates her new victim with the disease.

Larval mosquitoes feed on organic debris (with the exception of a few species that are predators). They use a pair of mouth brushes to strain out small aquatic organisms and particles of plant and animal material present in the water.

Seasonal Abundance

Mosquitoes may breed and develop any time from the beginning of spring to the first hard frost of fall. In general, populations are highest in summer and early fall. There may be from one to several generations of mosquitoes during a season depending on the species, the temperature, and the amount of rainfall.

When rainfall is abundant, many species can lay eggs continuously. Under ideal conditions with high temperatures, development can be completed in less than a week, resulting in large populations of flying adults.

Medical Importance

Worldwide, mosquitoes transmit many debilitating and fatal diseases, especially in tropical, developing countries. The most important of these is malaria, which has been on the increase in the last decade. In the United States, mosquitoes are primarily an annoyance, causing itching bites and welts that can become secondarily infected. Human mosquito-transmitted diseases remain relatively rare, due largely to modern pest control methods and disease detection. Encephalitis, among humans, and dog heartworm, among dogs, are the main diseases transmitted in the United States

Encephalitis. At least six types of mosquito-transmitted encephalitis occur in the United States. These are eastern equine encephalitis, western equine encephalitis, California encephalitis, St. Louis encephalitis, Venezuelan equine encephalitis, and La Crosse encephalitis. Each type is caused by a different virus or virus complex affecting the central nervous system. These viruses are normally transmitted by mosquitoes from birds or small mammals. Occasionally horses or humans are infected. Despite the small number of people infected annually by eastern equine encephalitis, it is considered a serious disease because it is often fatal.

Dog heartworm. This is a filarial parasitic disease transmitted by a number of different mosquitoes to dogs and, rarely, man. Once a problem only in coastal areas, dog heartworm is now found in every state in the United States. The nematodes, which lodge and grow in the heart tissue, can be fatal to dogs if left untreated.

There has been some concern about whether mosquitoes are capable of transmitting AIDS from an infected person to an uninfected person. Unlike encephalitis viruses and other mosquito-transmitted diseases, the HIV virus that causes AIDS is not able to survive inside the body of the mosquito. It has never been proved, and researchers say it's virtually impossible that a mosquito could transmit AIDS.

MONITORING AND THRESHOLDS

Introduction

Sampling and counting the mosquito population accomplishes a number of things. It helps determine whether mosquito control is necessary. It determines what growth stage the mosquitoes are in, providing information necessary to time control methods. It tells which mosquito species are present, especially important in areas of disease outbreaks. Finally, it helps to gauge how effective control efforts have been and when they need to be employed again.

Sampling should be done at least once a week, and more often during peak season. It is important to consistently sample the same sites each time. The numbers counted, the growth stage, and the species and sex should be needed when possible. All of this information gives an estimate of the population and must be compared with previous counts to determine whether the number of mosquitoes are increasing or decreasing.

A park manager can get an estimate of the number of mosquitoes in an area by counts of larvae or adults or both.

Larval Dipper Counts

Larval dippers can be purchased through biological supply houses or you can make your own. It is basically a shallow, plastic, enamel, or aluminum cup attached to a long handle. To collect floating mosquito larvae and pupae, depress one end of the dipper under the surface and quickly but smoothly scoop up larvae. If you move too quickly or cast a shadow on the surface, they will dive to the bottom.

The number of dips at each site will vary according to the size of the water body, but generally are in multiples of ten. Take five dips from open water and five from the water's edge, near vegetation if possible. Dipper inspections should be made weekly during the breeding season. Larvae can also breed in rainwater that has collected in containers such as buckets, garbage cans, canoes, tires, and animal watering troughs. To sample larvae in less accessible areas such as tree holes, use a large basting syringe to collect them. Empty them into a white pan for counting.

One advantage to sampling larvae is that the problem can be treated at the same time it is identified. When counting adult mosquitoes, the mosquitoes can be flying in from some distance away.

Adult Trapping

Trapping of adult mosquitoes gives information on the relative population size and the species composition.

Light traps are useful for monitoring certain species of mosquitoes. Not all species are attracted to lights. Different models of traps vary in the numbers, the species, and the proportion of males to females that they catch.

New Jersey light traps and CDC light traps (and their variations) are the traps most commonly used. Light traps are operated from dusk to dawn, powered either by electric line or a battery. Some traps are available with a photoelectric cell that turns the light on at dusk and off at dawn. When mosquitoes approach the light, they are blown by a small fan down through a funnel into a killing bag or jar.

The light trap should be hung about 6' off the ground in an open area near trees or shrubs but away from competing lights and buildings. Traps should be emptied each morning and the catch stored in a labeled box until it can be sorted and identified.

Since mosquitoes are attracted to carbon dioxide in the host's breath, some light traps are augmented with a one pound block of dry ice, wrapped in newspaper and hung next to the trap. The addition of dry ice also allows sampling on moonlit nights or in areas where bright lights may conflict with the light trap. And it allows daytime sampling of species that are active during the day or that are not attracted to lights.

Because some species are not attracted to light traps, they should be used in conjunction with other kinds of sampling methods. Monitoring for adult mosquitoes is an important part of the management of some mosquito-vectored diseases such as eastern equine encephalitis. The decision to use pesticides for mosquito supression is made only after intensive monitoring of the mosquito population in an area to determine if the species that vectors the disease to humans is present. The incidence of the disease in the wild animal population is monitored as a way to estimate the possibility of transmission to humans. Visitor education is also emphasized to alert people to the presence of the disease and how to go about protecting themselves.

Adult Landing/Biting Counts

Collecting mosquitoes as they land to bite is a convenient method of sampling biting populations. It involves rolling up a sleeve or pants leg and sitting quietly for a designated period of time, usually 10 minutes. During that time, each mosquito that lands on the leg or arm is collected with a battery or mouth-operated aspirator. It is important that you collect the landing mosquitoes for counting and identification and to ensure that you don't count the same individual again. Biting counts are best conducted from 30 minutes before sunset to 30 minutes after sunset (unless sampling day-biting species) by the same person each time.

The advantage to using landing counts as a sampling device is that you are counting only **biting** mosquitoes. The method does not collect male mosquitoes or species that do not actively bite people. It can also be used to count and collect daytime biters.

When sampling adult mosquitoes, sample all areas where mosquitoes may be a nuisance. Sample areas from which you have received complaints and near areas with high larval or pupal counts. Sample the same sites regularly, from one to seven nights a week. Adult mosquito information is most useful in gauging the extent of the mosquito problem, since it is the adults which transmit disease or create a nuisance.

Threshold/Action Population Level
The data from sampling and monitoring will be used to help decide at which infestation level to initiate management tactics. This decision level will be based on larval and adult counts, complaints from visitors, the potential for disease outbreaks, and the risk of the management tactics to other animals. For instance, in an area where there have been encephalitis cases, the risk is higher and the action level will be lower than in other areas.

The number and location of visitor or neighbor complaints should be plotted on a graph against the counts of immatures and adults for the same date and site. The amount of unacceptable complaints is the injury level. The graph should show the number of mosquitoes that correspond to the complaint injury level. This is the action level.

Action levels are different for each situation. In some areas, general annoyance does not occur until the number of female mosquitoes caught in light traps exceeds 25 per night. Other action levels that have been used are landing rates averaging more than 5 mosquitoes in 10 minutes and dipper counts averaging 5 larvae per dip. However, in most National Park Service locations, the action level would be higher than in a suburban neighborhood.

NON-CHEMICAL CONTROL OF MOSQUITOES

Introduction

The key factor in a mosquito integrated pest management program is determining whether or not control is necessary. This decision requires a regular mosquito sampling program to determine what species are present and in what numbers, and a set of action thresholds to determine if management tactics are necessary. If control is needed, then decisions have to be made on the best combination of tactics to suppress the mosquito population while affecting the environment as little as possible.

Normally, source reduction--eliminating or altering the water so that the mosquitoes cannot breed or complete their life cycle--is the first choice for control. If source reduction is impossible or incomplete, the next tactic to consider should be biological control of the larvae with predators, bacterial insecticides, or growth regulators. Visitor education also represents an essential part of a mosquito integrated pest management program at national parks. Interpretive displays can be used to explain the role of mosquitoes as a food source for animals such as bats, birds, and fish, and to help visitors understand that not all mosquitoes bite or carry disease. Personal protection through the use of proper clothing and repellents can be explained, as well as the avoidance of areas with high mosquito populations.

Source Reduction

The simple fact that all mosquito species require water to develop is the key to their control. No standing water means no mosquitoes. Source reduction is the first step in an integrated pest management program for mosquitoes. It is simply the use of mechanical methods to eliminate standing water. Source reduction involves filling, deepening, draining, ditching, managing water levels, maintaining shorelines, managing aquatic and inundated vegetation, and others. While these methods may prove to be more extensive and more expensive than some other controls, in most cases they need be done only once. Unfortunately, these methods will most likely require permitting from several agencies before they can be implemented. They are also not feasible in natural zones of a park.

Source reduction controls the immature mosquito stages--eggs, larvae, and pupae. Because these stages are concentrated in discreet bodies of water, they are much easier to control than are dispersed adult mosquitoes. Two water management tactics are ditching and ponding. That these would only be allowed in a developed zone. Ditching controls mosquitoes in two ways. In some cases water drains out of the potential breeding sites. In others, ditching allows fish access to the isolated pools where they prey upon the larvae and pupae. Ponding is another water management tactic that turns a temporary pool breeding mosquitoes into a permanent one capable of supporting fish and other mosquito predators. Ponding is accomplished by raising the water level, digging new pools, or through impoundment.

If standing water can't be eliminated, control of mosquito larvae in the water is the next step. This is best done with natural controls such as mosquitofish or biorational insecticides. The latter do not affect pupae and should not be used if this is the predominant life stage.

Biological Controls

Mosquito-Eating Fish

Fish are the most important predator of mosquito immatures. Mosquitoes are rarely a problem in a body of water that also contains fish. To use fish as a biocontrol agent the water must be deep enough and must have the right combination of environmental conditions to sustain fish. Introduced fish must have protection from native fish and other aquatic predators. The mosquitofish, *Gambusia affinis,* is often reared and released to control mosquitoes. However, this fish tends to outcompete native fish if not managed with care.

Bacterial Insecticides

Various commercial products containing *Bacillus thuringiensis* israelensis (*B.t.i.*) are available for treating bodies of water. This bacteria kills mosquito and blackfly larvae. It is nonhazardous to humans, other animals, fish, and predacious insects. *B.t.i.* is available as granules, slow release briquettes, or wettable powder. It can be applied by hand, with a backpack blower or granule spreader, or by aircraft.

Because the released bacterial spores must be ingested by the larvae, *B.t.i.* is not effective against eggs, pupae (which do not feed), or mature larvae that are ready to pupate and have stopped feeding.

Natural Enemies

Mosquito larvae are an important food for aquatic organisms. Large numbers fall prey to fish, insects, and spiders. Naturally-occurring bacteria, protozoa, fungi, and nematodes also kill mosquito larvae. Both bacteria and predatory fish have been used as biocontrol agents to control mosquito larvae. Adult mosquitoes are fed upon by birds, bats, frogs, lizards, spiders, and insects.

Other Controls

Screening of doors, windows, and vents is a time-honored method of keeping mosquitoes out of structures. Ordinary window screen of 16 x 16 or 14 x 18 meshes to the inch will keep out most mosquitoes. Campers can hang mosquito netting over cots, tent openings, picnic tables, etc. Long sleeves, long pants, hats, and veils give additional protection from mosquitoes.

Insect electrocuters, or "bug zappers," do not effectively control mosquitoes. Many mosquitoes are not attracted to the light. Tests in residential areas have shown that only a tiny percentage (usually less than 3%) of the insects killed are mosquitoes. Most are harmless gnats, moths, and beetles.

CHEMICAL CONTROL OF MOSQUITOES

Growth Regulators

Insect growth regulators such as methoprene do not kill the larvae but prevent them from developing into adults. Timing of application is important since only mature larvae are affected. Larvae that have already pupated will continue to develop into biting mosquitoes. Methoprene can be applied as slow-release briquettes, granulars, or ground or aerial spray. Most insect growth regulators do not harm other nontarget species.

Since methoprene does not kill the immatures, you will still collect larvae and pupae in dip counts. The only way to determine whether the treatment was effective is to rear the collected larvae and pupae and observe whether they develop into adults.

Larvaciding

Petroleum oils or specialized mineral oils can be applied to the water. The oil forms a thin film over the surface which suffocates eggs, larvae, and pupae. In the presence of wind, waves, or rain, the oil film breaks up and is less effective. Some oils are toxic to fish, other organisms, and aquatic plants. Various insecticides can be applied to the water as dusts, granulars, wettable powders, or emulsions. Consult your regional Integrated Pest Management Coordinator for specific recommendations for your area. Pesticides will likely kill other aquatic insects and may be harmful to fish, birds, and mammals.

Adulticiding

Adulticiding is space spraying for adult mosquitoes with insecticides. With an effective source reduction and larviciding program, adulticiding should not be necessary. It is generally the last resort in an integrated mosquito control program, since spraying of adult mosquitoes provides only temporary relief. It must be repeated frequently to intercept new mosquitoes moving into the area. Adulticiding may be the only feasible management strategy in a natural area where mosquitoes pose a public health risk, since source reduction is prohibited.

Most adulticiding is accomplished with a truck-mounted, ultra-low volume sprayer. Depending on the size of the area to be controlled, other application methods include backpack or power sprayer, mist blower, thermal fogger, or application from aircraft. Spraying is usually done in early evening when winds are less than 6 mph. Ground spraying is not possible in most natural areas.

Insect repellents

Personal insect repellents containing DEET applied to skin or clothing provide protection from biting. Another repellent, permethrin, may be used on clothing only. Jackets and tents impregnated with repellents are also available.

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Museum Pests

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for museum pests. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

A pest in a museum can cause far more damage than the same pest in a home or an office building. Carpet beetles in a stuffed bear, clothes moths in a native American headdress, or cigarette beetles in a herbarium can destroy irreplaceable artifacts. But while museum specimens must be protected, their sensitivity to chemical and environmental stresses means that standard pest control procedures are often unacceptable. Liquid pesticides may stain certain materials, heat treatment can damage paintings, dusts can abrade sensitive specimens. Caution is the byword in museums.

Each new pest problem must be analyzed on its own. Before taking any actions to control a pest, be sure that those actions will not themselves damage the museum's collections. Although the use of pesticides may at times be necessary, indiscriminate use or dependence on pesticides is unacceptable.

Any pest with chewing mouthparts is a risk to museum specimens. Carpet beetles, clothes moths, powderpost beetles, cockroaches, and others pose direct threats to specimens through feeding damage, feces, and excretions. Some pests pose indirect risks such as fires (rodents gnawing on wires) and secondary infestations (dead cluster flies in attics can attract carpet beetles).

IDENTIFICATION AND BIOLOGY

Any pest that infests houses, restaurants, or other buildings may at some time become a pest in a museum. Certain pests, however, repeatedly threaten museum collections. They can loosely be grouped into five categories:

(1) Fabric pests
(2) Wood pests
(3) Stored product pests
(4) Moisture pests
(5) General pests

The first step in solving any pest problem is to identify the pest and learn about its biology and habits. While it is impossible to discuss each pest in detail in this manual, the brief discussions below may help you understand a little about the habits of the pests most likely to infest museums or damage museum specimens.

Fabric Pests

Most insect damage to fabrics is caused by carpet beetles (in the family Dermestidae) or clothes moths (in the family Tineidae). The adults stage is seen most often seen since adults fly and some are attracted to lights and windows, but it is not the adult insects that do the damage. They feed outside on pollen or not at all. It is the larva or immature stage that feeds on fabric, fur, feathers, or virtually anything made of animal fibers.

Carpet Beetles

Immature carpet beetles feed on dried animal products such as wool, silk, felt, hair, fur, feathers, dead animals, and stuffed trophy heads. They do not feed on synthetic fabrics, but sometimes damage wool-synthetic blends or synthetics stained with urine, sweat, or food.

Carpet beetle larvae are repelled by light and are usually found burrowed deeply into infested material or in little-used drawers, cases, and storage bins. To grow, they molt and shed their skins. In heavy infestations, you may find large numbers of these light-colored shed skins. The adults are often seen crawling up walls and congregating on window ledges.

There are many species of carpet beetles. In addition, many common beetles resemble carpet beetles. Be sure to get the pest beetle properly identified so that you can zero in on the infested goods and likely harborage sites. Four species of carpet beetle are most likely to be found in museums.

Black carpet beetle (*Attagenus unicolor*) is the most abundant and destructive of the carpet beetles. The adult is 1/8"- 3/16" long, a solid dark brown or dull black color, and more elongate than carpet beetles described below. The larva is less than 1/4" long and carrot-shaped. It is covered with golden brown hairs and has a characteristic "tail" of long hairs at the rear end.

Varied carpet beetle (*Anthrenus verbasci*) is primarily a scavenger. It is common in the nests of birds, on dead animals, and in insect collections, but can damage woolens, carpets, wall hangings, hides, horns, and bone artifacts. Small populations often go unnoticed behind furniture or along baseboards feeding on accumulated lint, hair, food crumbs, dead insects, and other organic debris. The adult is about 1/8" long, oval to round, with splotches of white, yellow, and black on its back. The larva is tear-drop shaped and covered with rows of light brown hairs.

Common carpet beetle (*Anthrenus scrophulariae*) attacks carpets, woolens, and animal products such as feathers, furs, leather, silks, mounted museum specimens, and pressed plants. The adult is about 1/8" long with a band of orange scales down the middle of its back. The larva is reddish-brown and covered with brown or black hairs. A mature larva is active and moves rapidly.

Furniture carpet beetle (*Anthrenus flavipes*) attacks furniture (particularly old horsehair-stuffed furniture) and items made from wool, fur, feathers, silk, horns and tortoise shell. The adult is about 1/8" long, and is rounded and blackish with variable mottling of yellow and white scales on the back and yellow scales on the legs. The larva is difficult to tell from the common carpet beetle.

Clothes Moths

These are small, silvery-beige moths with a wing span of less than 1/2". They have narrow wings fringed with long hairs. Small grain and flour-infesting moths are often confused with clothes moths. However, clothes moths have different flying habits. They avoid light and are rarely seen flying. They prefer dark corners, closets, and storage areas, and usually remain out-of-sight.

The primary food of clothes moth larvae is soiled woolens, but they also feed on silk, felt, fur, feathers, and hairs. In museums they often damage woolen clothes (particularly old military uniforms), feather hats, dolls and toys, bristle brushes, weavings, and wall hangings.

The webbing clothes moth (*Tineola bisselliella*) and the casemaking clothes moth (*Tinea pellionella*) are the two most common clothes moths found in museums. The larvae are small white caterpillars with brown heads. They feed on the surface of the material infested. The webbing clothes moth produces feeding tunnels of silk and patches of silken webbing on the fabric's surface. The casemaking clothes moth is rarely seen since it constructs a cylindrical case of fabric which it carries around to hide and feed in. The color of the larva's case can help you locate infested materials.

Wood Pests

Materials made of wood are susceptible to attack by a number of wood- infesting pests. The culprits in museums are usually powderpost beetles or drywood termites. Both can severely damage valuable artifacts while remaining invisible to the untrained eye.

Powderpost Beetles

These are a group of beetles in the insect families Anobiidae (anobiid, furniture, and deathwatch beetles), Lyctidae (true powderpost beetles), and Bostrichidae (false powderpost beetles). The term "powderpost" comes from the fact that the larvae of these beetles feed on wood and, given enough time, can reduce it to a mass of fine powder.

Powderpost beetles spend months or years inside the wood in the larval stage. Their presence is only apparent when they emerge from the wood as adults, leaving pin hole openings, often called "shot holes," behind and piles of powdery frass below. Shot holes normally range in diameter from 1/32" to 1/8", depending on the species of beetle. If wood conditions are right, female beetles may lay their eggs and reinfest the wood, continuing the cycle for generations. Heavily-infested wood becomes riddled with holes and galleries packed with a dusty frass (wood that has passed through the digestive tract of the beetles). Both hardwood and softwood can be attacked by powderpost beetles, although lyctids only infest hardwoods.

Items in museums that can be infested by powderpost beetles include wooden artifacts, frames, furniture, tool handles, gun stocks, books, toys, bamboo, flooring, and structural timbers.

Drywood Termites

Unlike their cousins the subterranean termites, drywood termites establish colonies in dry, sound wood with low levels of moisture, and they do not require contact with the soil. They are primarily found in the coastal southern states, California, and Hawaii, but they are easily transported to northern states in lumber, furniture, and wooden artifacts.

Drywood termites attack wooden items of all kinds. The termites feed across the grain of the wood, excavating chambers which are connected by small tunnels. The galleries feel sandpaper-smooth. Dry, six-sided fecal pellets are found in piles where they have been kicked out of the chambers. The pellets may also be found in spider webs or in the galleries themselves.

A swarming flight of winged reproductive termites can occur anytime from spring to fall. Most drywood termites swarm at night, often flying to lights.

Stored Product Pests

Many museums include items made in part of seeds, nuts, grains, spices, dried fruits and vegetables, and other foods. A long list of pests, traditionally called "stored product pests" or "pantry pests," can infest items containing these foods. Probably the most common of such pests in museums are the cigarette beetle and the drugstore beetle.

Cigarette beetle (*Lasioderma serricorne*) is named for the fact that it is a pest of stored tobacco, but is also a serious pest of flax, spices, crude drugs, seeds, and, most importantly for museums, books and dried plants. This beetle has been called the "herbarium beetle" because of the damage it can cause to dried herbarium specimens. It has also been found infesting rodent bait.

The adult beetle is light brown, 1/8" long, and the head is bent downward so that the beetle has a distinctive "hump-backed" look. It is a good flier. The small larva is grub-shaped and whitish, with long hairs that make it appear "fuzzy." It has yellow- brown markings on the head.

Drugstore beetle (*Stegobium paniceum*) feeds on a wide variety of foods and spices (particularly paprika or red pepper). It is also a serious pest of books and manuscripts, has been reported "feeding on a mummy," and has been known to chew through tin foil and lead sheeting.

The adult beetle is very similar to the cigarette beetle. With careful examination through a magnifying lens, the drugstore beetle may be distinguished by its three- segmented antennal club. The larva, too, is similar, but does not appear as "fuzzy."

Moisture Pests

Moisture is not only a threat to museum specimens on its own, it may attract a number of moisture-loving pests that can do additional damage. The most important of such pests are the molds and insects in the order Psocoptera that feed on those molds.

Molds. Molds are fungi that can cause damage or disintegration of organic matter. Basically plants without roots, stems, leaves, or chlorophyll, molds occur nearly everywhere. When moisture and other environmental conditions are right, molds can appear and cause significant damage to wood, textiles, books, fabrics, insect specimens, and many other items in a collection. Their growth can be rapid under the right conditions.

It is important to realize that fungal spores, basically the "seeds" of the fungus, are practically everywhere. Whether molds attack suitable hosts in a museum depends almost exclusively on one factor: moisture. When moisture becomes a problem, molds will likely become a problem too.

Psocids. Although psocids are commonly called booklice, they are not related to parasites such as head lice or body lice. Booklice got that name because they often infest damp, moldy books. They feed on the mold growing on paper and in the starchy glue in the binding. Psocids also infest such items as dried plants in herbaria, insect collections, manuscripts, cardboard boxes, and furniture stuffed with flax, hemp, jute, or Spanish moss.

Psocids do not themselves cause damage. They become pests simply by their presence. However, their presence also indicates a moisture problem and the likely presence of damaging molds. They are tiny insects, less than 1/8" long, and range in color from clear to light grey or light brown. Most indoor psocids are wingless, looking a bit like a tiny termite.

General Pests

Any household pest may become a pest in a museum. Cockroaches, rodents, silverfish, ants, and other common pests can invade and infest a museum as well as a house or other structure. The biology and ecology of these pests are covered in detail in other modules of this Integrated Pest Management Information Manual, and will not be repeated here.

MONITORING AND THRESHOLDS FOR MUSEUM PESTS

Visual

Regular and scheduled inspections of all specimens on display and all collections in storage can prevent pest infestations from building up undetected. Specimens on display should be checked monthly. All collections in storage should be opened and examined at least twice a year.

Monitors should use a bright flashlight during inspections and look for live adults and larvae and the presence of shed larval skins or feces. The presence of feeding debris or frass around or below specimens is an indication of infestation. So are exit holes, feeding holes, hair falling from fur or pelts, mats of fibers, silken feeding tubes or cases, or moth or beetle pupae. A hand lens can be used to examine for eggs if an infestation is suspected.

Window sills and the inside of ceiling light fixtures should be checked on a regular basis as many pests fly or crawl to light. The immediate display area should also be examined. Pests may be found behind baseboards, under furniture, behind moldings, in cracks in floors, behind radiators, or in air ducts.

Traps

Small **sticky traps** should be placed in hidden areas throughout the facility and inside specimen cases, particularly in high-risk areas. These traps should be checked at each visit, any pests identified and recorded, and the traps replaced as necessary.

Pheromone traps are one of the most valuable new tools for pest management in museums. Pheromones are the natural scents insects use to communicate with each other. Certain pests can be strongly attracted to the traps from the surrounding area, providing an extremely effective early warning system of pest presence.

Pheromone traps are only available for certain insects. Traps useful in museum settings include those for cigarette beetles, drug store beetles, Indianmeal moths, and warehouse beetles (*Trogoderma*). Others are being developed and may be available soon.

Insect electrocutors are useful for detecting and controlling flying insects. They emit ultraviolet light (black light) that attracts flying insects, particularly flies and moths. The insects are drawn into the trap and electrocuted or fall onto a glue board. These traps must be checked and emptied periodically or the dead insects will themselves attract dermestid beetles and other scavengers.

Recordkeeping

Careful records of inspection results, trap catches, etc. will help identify seasonal risk factors and areas with a high frequency of problems.

Threshold Levels

Because of the sensitivity of museum collections, for most pests in the immediate museum area the action level will be one live specimen. Presence of live adults or larvae indicate ongoing infestations which should be investigated immediately and treated as necessary. Shed larval skins and feeding damage may have resulted from old infestations, but in regularly monitored collections, these should be regarded as an indication of an active infestation. Thus, it is vitally important to maintain careful monitoring records.

NON-CHEMICAL MANAGEMENT OF MUSEUM PESTS

Introduction

Pest management in a museum is especially tricky because without careful management the cure could cause as much damage as the pest. Museum specimens by their nature are rare and valuable. They are often delicate and liable to stain, warp, or simply fall apart if control procedures are too aggressive.

Ideally, the focus of integrated pest management in a museum will be on habitat modification and exclusion to **prevent** pests rather than on control methods to eliminate them.

Safe and successful museum pest management requires an integrated pest management approach, combining careful and frequent monitoring of pest levels and conditions with a combination of tools, procedures, and strategies. The tactics chosen for a particular pest problem should be adapted to the conditions in a museum. Anticipate the consequences of each tactic. When deciding from among them, choose the combination of tactics least likely to put specimens and visitors at risk.

Nonchemical management tactics include cultural controls (temperature and humidity control, sanitation, lighting), pest-proofing (pest-proof containers or display cases, screening and caulking, etc.), trapping (mechanical, sticky, pheromone, and light traps), vacuuming, freezing or heating infested specimens, and, in rare cases, "radiation" such as microwave ovens and gamma irradiators.

Cultural Control

Sanitation plays an important role in the attractiveness to pests of a museum area. Poor sanitation--food debris, grease, loose hairs, and the like--in and around specimens, storage areas, and in cracks and crevices in floors and furniture attracts and holds pests. Good sanitation, particularly regular mopping, washing, and vacuuming, removes potential foods and even newly-arrived foraging pests.

Controlling lighting can also reduce the attractiveness of an area to pests. Minimize exterior lighting. Bright lights shining through doorways and windows can attract insects to the museum area. Light shields, curtains, and closed doors can reduce the numbers of flying insects attracted to the museum.

Temperature and humidity control also can affect pest populations. Lowered humidity and, to a lesser extent, lowered temperatures reduce the chance of infestation and slow down the growth of existing pest populations. For some pests, such as psocids, reducing humidity can be all that is required to eliminate a pest problem.

Pest-Proofing

The most effective way to prevent damage from dermestid beetles, clothes moths, and many other museum pests is to prevent establishment of infestations in the first place. Preparation of specimens should take place in areas other than collection rooms. All incoming specimens should be examined carefully for damage and live insects, and records kept. Incoming specimens showing signs of infestation should be isolated and disinfested. Contact your regional curator before undertaking any control measures on unfamiliar specimens.

Windows in areas where specimens are kept should be tightly screened or kept closed at all times to prevent pest entry. Caulk or otherwise seal cracks and holes in walls and floors, holes around pipes and other utility lines, and other points of pest entry. Install door sweeps where necessary. Air vents and hot air registers can be equipped with filters to trap potential incoming pests. Filters should be changed on a regular basis.

Adult dermestids and other museum pests feed on pollen and nectar, so decorative cut flowers should be kept out of specimen areas to reduce the chance of accidental infestation. Those specimens at high risk of insect damage should be kept in insect-proof cases and examined on a regular basis.

Trapping

Besides their monitoring function (as mentioned earlier), traps may also be used to control pests. Snap traps and glue boards are often used against rats and mice. Pheromone traps are also a good supplemental control tactic for certain pests, particularly in removing the last few individuals left in the area. Likewise, insect electrocutors and sticky traps can supplement other control measures.

Cold Treatment

Some infested museum specimens can be disinfested by freezing them in a large commercial freezer that can reach temperatures of 0 F or lower. Herbarium specimens, books, mammal and bird collections, as well as various ethnographic materials, have been successfully frozen for insect control. For example, you can kill lyctid beetles by holding the specimens at 0F for at least 48 hours, although four days is preferable. Most other pests can be destroyed by the same regime. Books are commonly disinfested by wrapping them tightly in plastic and freezing them for one to two weeks. Note that freezing poses a significant risk of damage to certain woods, bone, lacquers, some painted surfaces, and leather. Check with your regional curator if in doubt. Low, but above-freezing temperatures, usually 40 F-42F, can be used to protect items in storage. The best example is low-temperature storage of furs and costumes.

Heat Treatment

Small items can be heated in an oven to kill infesting pests. Larger items may require a commercial kiln. Powderpost beetle larvae and eggs will be destroyed if the internal temperature of the wood is held at 120F for two hours. Holding a specimen at a temperature of 130F for three hours will kill any insect. However, this level of heat may damage veneers or the finish of specimens, warp lumber, or melt glues. Check with your regional curator if in doubt.

Microwaving

Microwaving as a pest control method is mostly an experimental technique at this time. However, it can be an option as long as the treatment will not damage the item being disinfested. For example, microwave ovens have been used to kill cockroaches, silverfish, and psocids inside books. The average infested book is microwaved on high for 20-30 seconds. Longer times risk damage to the glues and bindings of many books.

This method is safe for most hardback books printed after 1950 and high- quality soft-cover books with sewn bindings. Do not use this method on valuable old editions, older books with metallic dyes, inexpensive soft-covers (it will melt the glue), or books bound in leather.

Miscellaneous

Cedar wood chests are often recommended to protect fabrics from clothes moths and carpet beetles. However, only freshly cut cedar wood is toxic or repellent to fabric pests, and then only in an air-tight container. By the time the wood is two years old, there is no toxic effect left. (Of course, a tightly sealed box of any material will usually keep pests out.)

CHEMICAL CONTROL OF MUSEUM PESTS

Pesticides used in museum pest control are generally some of the same products used for household or other structural pest control. Most pesticide products are not specifically labeled for use in museums. If the product is not so labeled, be sure it is labeled for use in similar sites such as public buildings, institutional settings, etc.

Museums are a good site in which to use nonconventional pesticides such as repellents and insect growth regulators (IGRs) for controlling cockroaches, cigarette beetles, and certain other stored product pests. Cockroach and ant bait stations are an excellent choice for these pests since they pose no risk to museum collections.

When using any pesticide for general pest control (cockroaches, silverfish, ants, etc.), avoid direct treatment of museum specimens whenever possible. Instead, treat cracks and crevices, wall voids, and perhaps the legs of display cases rather than inside the cases themselves.

Paradichlorobenzene and naphthalene are commonly used as repellents in museum cases. These materials do not eliminate infestations, but may be useful in preventing them. Paradichlorobenzene and naphthalene may cause damage to certain plastics (bakelite, for example), and may soften and shrink resins, adhesives, and paints. Organic gas filters should be installed on the sides of cabinets to absorb fumes and replaced when the odor is detected in the room.

Specimen cases are sometimes treated with insecticidal dusts. This treatment poses some risk of abrasion to specimens stored in the case, and some risk to curators later working with the specimens. Such treatments should be done with care.

Treating Infested Materials

If nonchemical treatment of infested materials is not practical, some materials can be treated with standard insecticides. However, in most situations, infested museum specimens should be fumigated. **Fumigation is hazardous and it requires professional training to do it safely and effectively.** Fumigation of museum specimens is normally conducted in special fumigation chambers, vaults, or "bubbles." Some fumigation is done under tarpaulins. In severe and extensive infestations, an entire building may have to be "tented" and fumigated.

There are a number of different fumigants to choose from. The choice will depend mostly on the objects and materials to be fumigated, since different fumigants are best suited for certain jobs. Some fumigants cannot be used on certain materials because they may react with them (for example, methyl bromide may react with rubber goods). The most commonly used fumigants for museum specimens are methyl bromide, sulfuryl fluoride, ethylene oxide, and carbon dioxide.

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Rats

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for rats. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Rats have caused more economic loss and more human suffering than any other vertebrate pest. From plague epidemics (the "Black Death" of Europe) to rat bites of inner- city children, from gnawing electrical wires in an attic to feeding on stored food in a warehouse, rats are a critical pest of humankind.

Rats have adapted well to living around people. So well, in fact, that rats are commonly called "domestic" rodents. They live and breed inside buildings and granaries, in city sewers and attics, in agricultural fields and warehouses, in ships and under concrete slabs. Although adapted to people, however, rats are wary. Hundreds can be living in, under, and around a complex of buildings with few people in the area aware of their existence.

Successful management of pest rats is not easy. It requires an integrated approach based on a good understanding of the biology and habits of pest rats, that combines effective inspection and monitoring with intelligent use of control tactics.

IDENTIFICATION AND BIOLOGY OF RATS

Major Pest Species of Rats

When we speak of rats, we are speaking of many different species, some pests, some not. In the United States, the two most important pest rats are the Norway rat and the roof rat. The two species look similar but are noticeably different both in appearance and in habits.

Norway rat (*Rattus norvegicus* Berkenhout). The most common pest rat in the United States, the Norway rat is also called the brown rat, sewer rat, or wharf rat. The adult is a large, stocky rat, from 12"-18" from the nose to the tip of the tail, weighing 12-16 ounces. In contrast to the roof rat, the Norway rat's tail is shorter than its head plus its body, its ears are small (cannot be pulled down to reach its eyes) and covered with hairs, and its nose is blunt.

Roof rat (*Rattus* (L)). A common rat of coastal areas, the roof rat is also called the black rat or the ship rat. The adult has a slender body, weighs 5-9 ounces, and measures 13"-18" from the nose to the tip of the tail. In contrast to the Norway rat, its tail is longer than its head plus its body, its ears are large (can be pulled down to reach its eyes) and nearly hairless, and its nose is pointed.

Geographic Distribution

Norway rat. Found in every state in the United States and common throughout much of populated North America, Norway rats absent from sparsely-inhabited areas, particularly in the western states.

Roof rat. Roof rats occur mainly in coastal areas, including California, Oregon, Washington, the Gulf states, and the Southeast and mid-Atlantic states.

Life Cycle

Rats are born hairless, with eyes and ears closed. They don't become furry and "rat-like" until they're about two weeks old. They begin eating solid food at three weeks and imitate their mother to learn to forage, escape, and watch for danger. They are weaned at four or five weeks. Rats become independent of their mother and ready to start families of their own, at three months.

Norway rat. Norway rats breed at any time during the year, but more frequently in warm months. Gestation lasts 22-24 days. The size of the litter is usually 8-10 pups. There are 3-4 litters per year. Lifespan in the wild is usually about 6 months.

Roof rat. Root rats may breed throughout the year, but more commonly in warm months. Gestation lasts 20-22 days. The size of the litter usually 4-8 pups. A female may breed at 2-5 months of age and have an average of 5.4 litters per year. Life span in the wild is usually 9-12 months.

Seasonal Abundance

Outdoor rat populations tend to peak in summer to early fall. They tend to be at their lowest levels in late winter to early spring. Indoors, rat populations may remain at the same levels throughout the year, limited only by periodic shortages of food, water, or nesting sites.

Nests

Rats are social. They live together in colonies with well-defined territories and a social hierarchy or "pecking order." Norway rats and roof rats prefer different types of nest sites.

Norway rat. Outdoors, Norway rats usually dig shallow burrows in the ground. The burrows are generally less than 18" deep and 3'long, with a central nest. The main burrow opening is 2"-4" in diameter. Hidden "bolt holes" are used for emergency escapes. Indoors, Norway rats will nest inside walls, underneath equipment, in cluttered storage areas, and similar sites, usually on the lower floors of a building.

Roof rat. The roof rat commonly nests above ground in trees, vine-covered fences, stacked lumber and woodpiles, and overgrown landscaping. Roof rats will sometimes nest in burrows if above-ground sites are limited. Indoors, roof rats prefer to nest in the upper levels of buildings in attics and ceiling and attic voids near the roof line. This species seems to be less dependent on man than the Norway rat and may live in forests far from human habitation, especially in warm areas.

Both species also nest in sewers and storm drains.

Range
Rats commonly travel 100'-150' from their nests looking for food and water and patrolling their territory. They may have several "hotel" nest sites in an area and will move from home base to spend several days in one of these secondary nest sites.

Responses to Environmental Factors
Rat abundance is dependent on availability of food, water, and shelter. They need about an ounce of food and 1/2 fluid ounce of water daily, although the roof rat can get by on less. Both prefer to nest where water is easily available.

While the Norway rat prefers to feed on protein foods like meat, fish, insects, and pet food, the roof rat prefers a more vegetarian diet. It feeds on fruits, nuts, seeds, berries, vegetables, and tree bark. But the roof rat will also feed on garbage, pet food, and meat if it is readily available. Rats often cache or hoard food in hidden areas for use when food supplies run short.

Rats are wary of anything new that appears in their territory. This fear of new objects can make baiting and trapping difficult since rats will at first avoid baits, bait stations, and traps, and may come to associate them with danger as a management program proceeds.

When Norway and roof rats are found together, Norway rats will usually, but not always, displace roof rats.

Medical Importance
Rats have always been of medical importance due to their transmission of human diseases.

Direct effects. Rat bites, particularly in urban areas, may be a serious health problem. An estimated 14,000-24,000 bites to humans occur each year. Infants and helpless adults (unconscious, invalid, and elderly) are subject to attack by rats. A small percentage of those bitten develop rat-bite fever, a bacterial disease carried in the teeth and gums of many rats. All rat bites should receive medical attention.

Rats can spread *Salmonella* food poisoning, Weils disease (leptospirosis), trichinosis, and other diseases directly through contamination of food and water with their urine and feces.

Indirect effects. Rats may indirectly spread a number of serious human diseases by way of fleas and mites, most notably plague and murine (scrub) typhus fever. (See Pratt et al. [1986] for a complete discussion of rat-borne diseases).

Outbreaks of rat-associated diseases. Some of the diseases listed above can be fatal to humans. If disease transmission is suspected in your areas, contact your National Park Service Public Health Service representative.

Rabies. Rats have never been found to be infected with rabies in nature, and rabies transmission has never been documented in the United States. The United States Public Health Service recommends **against** anti-rabies treatment in the case of rat bite.

MONITORING AND THRESHOLDS FOR RATS

Periodic surveys of buildings and grounds can reveal the existence of rat infestation. Inspection visits should be made every other week and increased or decreased according to the severity of the problem. Evening inspection using a powerful flashlight is the best way to see rats, but there are many signs of rat infestation besides the animals themselves. Rat sounds, droppings, burrows, urine stains, smudge marks, runways, tracks, gnawing damage, nests, food caches, pet excitement, and rat odors are all signs of rat activity.

Sounds. Squeaks, gnawing sounds, clawing, and scrambling in walls are typical sounds of a rat infestation.

Droppings. A single rat may produce 50 droppings daily. Norway rat droppings are larger (3/4") than roof rat droppings (1/2"). Determine if an area is currently infested by sweeping up old droppings, then reinspect after a week. Fresh droppings have a putty- like texture; old droppings crumble easily.

Burrows. Estimates of relative abundance in a limited area can be made by counting, mapping, and loosely plugging burrow entrances on a weekly basis. Burrows which are reopened the following week are active.

Nests. Roof rat nests are often visible in attics, or they may be found when vegetation is trimmed.

Urine Stains. Under ultraviolet light (blacklight), rat urine will glow blue-white.

Runways. Rats regularly travel the same routes. Outdoor runways appear as beaten paths in the ground. Grass will be worn down.

Smudge Marks. Oil and grease that rub off a rat's fur build up on well-used runways.

Tracks. An adult rat's footprint is about 3/4" long. Rats may also leave a drag line (from their tail) in the middle of their tracks. A "tracking patch" can help determine the location and extent of rat activity. Place a light dusting of clay, unscented baby powder, or powdered limestone in suspected runways and near rat signs. Typical patch sizes range from 12"x4" to 6"x18". Examine the patch for tracks at regular intervals.

Gnawings. Rats constantly gnaw on hard surfaces. Gnawed holes may be 2" or more in diameter.

Food Caches. Rats may store surprisingly large quantities of food in protected areas.

Pet excitement. Cats and dogs often probe an area of floor or wall where rats are active, particularly if the rats only recently invaded.

Odor. Heavy infestations have a distinctive odor. Experienced pest managers can smell the difference between a rat and a mouse infestation.

Learn to differentiate between fresh rat sign and old sign which may indicate old (non- active) infestation.

Evaluation of population size. Rat signs may be interpreted visually as follows.

Rat-free area or low rat population: no sign of rat presence.

Medium population: old droppings and gnawing common, one or more rats seen by flashlight at night, none during the day. Each rat seen at night usually indicates 10 or more elsewhere.

High population: fresh droppings, tracks, gnawing evident, three or more rats seen at night, one or more in daylight.

Estimates of rats present can also be made by placing premeasured, ground, nontoxic cereal bait in various locations to determined how much is eaten each night. Double the amount each night until the amount taken in one night levels off. Divide the amount by 1/2 oz. This will provide a very rough estimate of the minimum number of rats present.

In most circumstances the injury (threshold) level is one rat as determined by rat sighting or sign. The action level is one rat for population reduction programs and zero rats for prevention programs.

NON-CHEMICAL CONTROL OF RATS

Successful rat management programs use a combination of tools, procedures, and strategies. Some are lethal to the rat, some are not. Lethal procedures include the use of rodenticides, snap traps, and glue boards to quickly reduce a population. Nonlethal procedures include improving sanitation, reducing harborage, and rat-proofing buildings. Long- term, the most important tactics for reducing rat problems are in this second, nonlethal category, because the procedures reduce the environment's capacity to support rats or block the rats' access to buildings.

Before a park manager can decide what combination of strategies would be best for a particular situation, he or she needs to determine where the rats are nesting and feeding, locate their travel routes, and determine the extent of the infestation. This information is obtained through inspection and regular monitoring.

Improved Sanitation

Rats are attracted by food spills, open garbage, and food stored in accessible sites. Baiting and trapping programs often fail because the bait can't compete with the rats' regular food. Reducing the rats' food will reduce the rat-carrying capacity of the site, as well as making lethal control programs more effective.

In urban settings, rats feed largely on garbage. Regular trash pickups at the end of each day, rather then storing trash overnight, and the use of rat-proof trash containers are relatively simple methods of reducing rat food sources. Damaged dumpsters and containers should be repaired or replaced and should always be kept closed overnight.

Pet food dishes and water dishes should not be left full overnight. Bird seed is a favorite rat food. Bird feeders should be equipped with seed catchers, or the dropped seed should be cleaned up every evening.

Food in warehouse-style storage should be rotated properly-first in, first out. Food should be stored on pallets, not on the ground, and there should be about 2' of space between pallets and the side walls to permit inspection.

Harborage Reduction
Landscaping should not include thick hedges or bushes which obscure the ground. Ground covers such as ivy, which provide cover or runs for rats, should not be planted adjacent to buildings. High grass, weeds, wood piles, and construction debris should not be permitted near foundation walls. Dumpsters and outside garbage containers should sit on a paved or concrete pad. Indoors, reduce clutter in rarely-used rooms and organize storage areas.

Rat-Proofing
Building rats out of a structure, and keeping them out, is called rat-proofing.

Block openings around water and sewer pipes, utility lines, and air vents.
Install metal kick plates or sweeps on doors and metal jambs on windows and doors.
Screen air vents.
Seal any cracks or holes in foundations (above-and below-grade) and exterior walls.
Repair damaged roof soffits and seal any openings to the roof.
Repair any gnaw holes after stuffing them with steel or copper wool.
Equip floor drains with sturdy metal grates.

> In roof rat areas, cables, trees, and pipes leading to or touching a structure should be rat-proofed with galvanized metal barriers.

Trapping
The snap trap is an effective method of killing rats when used correctly. Traps are especially useful when you wish to avoid the use of poisons, to eliminate bait shy or bait resistant rats, to avoid odors from dead rats in inaccessible places, or to collect live rats for ectoparasite or resistance screening. The best traps are those with expanded triggers (treadles) set for a light touch. Set the traps along runways with the trigger towards the wall, or tie the traps to pipes or rafters or wherever droppings, gnawing, grease marks, and other evidence of activity is found.

The number one mistake in using traps is not using enough. (See Environmental Protection Agency [1991] for information on trapping).

Another way to trap rats is with glue boards. Glue boards are used much like snap traps. Secure the glue board with a nail or wire so it can't be dragged away. Be aware that some people may protest the use of glue boards as inhumane, since the rat may struggle for some time.

Natural Enemies

Rats may be preyed upon by many other animals including dogs, cats, weasels, snakes, and owls. Rats are susceptible to a variety of diseases and parasites. Some natural enemies ranging from ferrets to bacterial toxins have been used in the past with varying degrees of success in rat control programs.

In abnormally crowded conditions or other stress situations, rats may display aggressive behavior toward each other, including cannibalism and abandonment of young.

CHEMICAL CONTROL OF RATS

Rodenticides are commonly used to provide rapid reduction of rat populations. There are three major formulations of rodenticide: toxic baits, water baits, and tracking powders. Fumigants are also used occasionally to fumigate burrow systems.

Toxic baits. These combine a poison with a food bait attractive to rats. Today, most baits are obtained ready-made as extruded pellets, or in a dry meal, or molded into paraffin blocks for wet sites. Some baits kill rats in a single feeding, some require that a rat feed a number of times. Some are anticoagulants (causing rats to bleed to death), some affect respiration, and others work by entirely different modes of action. They range in toxicity to people from very toxic to slightly toxic. Be sure to read the label and supporting information that comes with each product to ensure safe use.

Every rodenticide has a warning on the label to place the bait "in locations not accessible to children, pets, wildlife, and domestic animals, or in tamper-proof bait boxes." What qualifies as a safe, inaccessible area needs to be determined on a case-by-case basis.

If you believe there is a risk to children or nontarget animals, the bait should be placed inside a tamper-proof bait box. A bait box is tamper-proof if a child or a pet cannot get to the bait inside. It is usually made of metal or heavy plastic. But a bait box is not truly tamper-proof unless it can be secured to the floor, wall, or ground.

In parks, there is the additional problem that there may be nontarget rodents that can find their way into bait stations (or traps, for that matter). Be sure to survey for these and adapt your management tactics to avoid harming them.

Water baits. These specially-formulated rodenticides are mixed with water and dispensed from "chick-founts," or custom toxic-water dispensers. Since rats drink daily, water baits are effective when free water is in short supply. Water baits are less effective against roof rats. Be sure to only use water baits where no other animals or children can get to them.

Tracking powders. These are rodenticides mixed with a talc or powdery clay and applied into areas where rats live and travel. The powder sticks to the rats' feet and fur and is swallowed when the rats groom themselves. Tracking powders are effective even where food and water are plentiful.

The rodenticide in tracking powders is 5 to 40 times more concentrated than in baits. Avoid applying tracking powder where the powder could become airborne and drift into nontarget areas, or where other nontarget animals may come in contact with it.

Fumigants. Several fumigants are available for burrow fumigation. Most are extremely hazardous and should only be used by experienced professionals. National Park Service policy for rat management emphasizes rodent-proofing rather than the use of rodenticides. Consult your regional Integrated Pest Management coordinator when considering their use.

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Silverfish

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for silverfish. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

IDENTIFICATION AND BIOLOGY OF SILVERFISH

The term silverfish is used for the Thysanura and for any of the species within the order. Thysanurans have a distinct carrot shaped body, short legs, long slender antennae and three tail-like appendages (anal cerci) at the end of the body. Silverfish are wingless with scale covered bodies which are about 1/2" long. Nymphs resemble adults.

There are thirteen species of silverfish (Thysanura) in the United States. Mallis (1982) lists six species which may be pestiferous: they are the firebrat, *Thermobia domestica* (Packard); *T. campbelli* (Barhart) (no common name), the silverfish, *Lepisma sacchrina* L.; the fourlined silverfish, *Ctenolepisma quadriseriata* (Luccas); the gray silverfish (also called the giant silverfish), *C. longicaudata* Escherich (*C. urbana* in some texts); and *Acrotelsa collaris* (Fab.) (no common name). See page 45 of U.S. Department of Health, Education, and Welfare (1967) for pictorial keys to common species. See also Mallis and Caur (1982) for detailed descriptions of pest species. Brief descriptions of the habitats and life cycles of the six pest species are given below.

Firebrat. This insect is found throughout the world in warm climates. Found indoors in the United States in heated buildings, it is occasionally seen outdoors in the West and Southwest. Firebrats occur around ovens, bakeries, and other warm areas, as they prefer temperatures above 90F. Firebrats may become serious pests in bakeries and in areas where starches are stored at warm temperatures, such as in warehouses.

The females deposit eggs in crevices. The number of eggs per batch ranges from 1 to nearly 200, with an average of 50, but stressed females lay fewer eggs. Eggs will hatch in 12-13 days under optimum conditions. Newly hatched nymphs are 1/8" long, white, and scaleless. Development is rapid, with only 1 day spent in the first instar and more time passing between successive instars, although the later instars may last up to two weeks. A firebrat may pass through 45 to 60 instars during its lifetime. The nymphs resemble adults. Females produce one batch of eggs per instar beginning at about the 12th week but can begin to oviposit at six weeks at temperatures of 90-106F. Firebrats can live up to two years at warm (90-98F) temperatures.

Firebrats can be killed when exposed to temperatures above 120 F for one hour or more. Temperatures above 112F and below 32 F kill nymphs. This can be an effective way to manage firebrats if it possible to elevate or reduce temperatures to these levels (Olkowski 1991).

***T. campbelli*:** This species is found indoors in libraries. Little is known about its habits, but its life cycle resembles that of the firebrat.

Silverfish. This pest is common indoors on the East Coast, and is also found indoors in the Midwest and Pacific Coast. It is found indoors in warm, humid areas such as basements.

The eggs are deposited in crevices or under objects singly or in groups of 2 or 3. The female deposits 1-3 eggs per day or at irregular intervals of up to several weeks depending upon availability of food. Eggs hatch in 43 days at 72F and in 19 days at 90F. Females may reproduce at 3-4 months of age. Nymphs are 1/8" in size and scaleless when hatched. The first instar lasts 7-10 days; successive instars are 2- 3 weeks long. Scales develop in the third instar. Adults may live up to 3 1/2 years, but most live 2 years under favorable conditions (72-80F, relative humidity of 75%- 97%). Silverfish may pass through up to 59 instars in their lifetimes.

Fourlined silverfish. This species is common on the East and West Coasts and in the Midwest. It lives indoors, often infesting attics, particularly if the roof is made of wooden shingles. It may be found outdoors in summer. Its life cycle is similar to that of the silverfish but not as limited by temperature and moisture.

Gray silverfish. This species occurs indoors in the South, Midwest, and southern California. It prefers drier areas than common silverfish, such as crawl spaces and attics, but may occur around water pipes in bathrooms. It deposits its eggs in cracks in groups of 2-20. They hatch in about 60 days at room temperature. The nymphs are scaleless when hatched; scales appear in the fourth instar, and sexual maturity is reached in 2-3 years. This species may live up to 5 years.

A. collaris: This species was recently introduced into Florida, probably from the tropics. Little is known of its life cycle but it may resemble that of silverfish.

Feeding habits of silverfish species are very similar. Once a source of food is located, silverfish remain in the vicinity. Silverfish feed on human foods, especially those containing starch or flour, as well as on paper, especially glaze-coated paper. They eat sizing on paper, as well as glue and paste. They may feed on wallpaper or the paste behind it, causing the wallpaper to become detached from the wall.

Materials such as writing paper, tissue, onion skin paper, and cellophane are preferred, as well as cotton, rayon, and other vegetable textiles. Newsprint, brown wrapping paper, and most cardboards are seldom eaten. Silverfish feed on bound volumes for the paper, the starch and sizing in the bindings, and the linen in some bookcovers. Stored papers, books, and other printed materials are especially susceptible. Sizing and glue are main attractants, especially if humidity is high. Silverfish seldom feed on wool and other animal based textiles. Cereals may become infested due to the insects' preference for starches and flour. Enzymes and cellulose-digesting bacteria in the gut break down cellulose in paper or other wood products. Silverfish can live for nearly a year without feeding.

Temperature is the most important factor influencing the thysanurans. Low temperatures result in high mortalities, especially among nymphs. Mallis (1982) reports that temperatures below freezing or above 112F result in 100% mortality in firebrat nymphs. Similar ranges can be expected for other species. Low relative humidities may reduce population growth or eliminate silverfish. In heated buildings, only food availability limits silverfish populations, and their numbers vary little throughout the year. Silverfish may enter buildings by way of boxes, books, or other materials carried inside.

Thysanurans are primarily important as archival pests although they may infest foodstuffs. Individuals are long-lived and reproduction rate is moderate, so populations grow slowly. Large populations can cause severe damage to paper and paper products.

MONITORING AND THRESHOLDS

Monitoring

Monitoring is best performed by detecting damage caused by silverfish (Mallis 1982). Book bindings will show minute scrapings. The sizing of paper will be removed in irregular fashion, and the edges of paper will appear notched. In cases of high populations irregular holes will be eaten directly through paper. Other signs of feeding include feces, scales, and small yellow stains.

Active infestations can be detected by observing the small, dark feces, which are visible to the eye as well as scales, which are visible through a hand lens (Mallis and Caur 1982). In addition the feeding activities of silverfish can be observed by coating a piece of paper with a thin layer of flour paste and placing it in an area suspected of harboring silverfish. If silverfish are present, the paper will show small feeding marks.

Two kinds of traps have been used to confirm the presence of silverfish. The first uses a small jar coated with flour on the inside and tape on the outside to provide a climbing surface. Jars should be placed in areas of suspected silverfish infestations and regularly inspected for silverfish, which will climb in the jar and become trapped (Mallis and Caur 1982). Conventional sticky traps such as those used for monitoring cockroach populations can also be used for detecting silverfish (Olkowski et al. 1991).

Thresholds

Due to their small size and reclusive nature, silverfish are seldom seen. However, their damage can be significant if populations are high. The decision to intervene should be based on the amount of damage associated with the silverfish and the confirmation of an active infestation by the methods described above.

NON-CHEMICAL CONTROL OF SILVERFISH

Warm temperatures and high relative humidities favor most silverfish species. Controlling or eliminating moisture in areas infested with silverfish can reduce populations. Air conditioners or dehumidifiers placed in rooms where documents and books are stored can help to reduce humidity and temperature. Lower temperatures may also slow population growth by reducing rates of development and reproduction in silverfish.

Silverfish found in books and documents can be killed by exposure to microwave radiation. Olkowski et al. (1991) recommend placing books in a microwave oven for a period of 30 to 60 seconds to kill silverfish. Caution should be used with books or documents containing color plates or in fragile condition.

Sealing cracks and crevices where silverfish hide and breed also reduces populations by reducing suitable habitat. If sealing or caulking is not possible, then cracks and crevices (particularly around bookcases) should be regularly vacuumed to remove silverfish. Good sanitation practices should be followed. All valuable paper products, books, and documents should be placed in tightly sealed containers and cabinets. If this is not possible, access to potential food sources should be limited by removal of food and harborage such as empty cardboard boxes and other waste paper.

Natural Enemies

There is no information in the literature on natural enemies of the Thysanura.

CHEMICAL CONTROL OF SILVERFISH

Several pesticides are recommended for use on silverfish in Park Service areas. Pesticides for silverfish should be applied in the same manner and with the same thoroughness as for cockroaches. Boric acid should be spread thinly in areas where silverfish are active. Dusts and powder formulations should be applied only in cracks, crevices, attics, and other storage areas where park visitors will not regularly come into contact with the pesticide (Olkowski et al. 1991). Consult your regional Integrated Pest Management coordinator to determine which chemicals and application techniques are best suited for your area.

SUMMARY

Regularly monitor high risk areas by looking for damage, droppings, scales, or insects or by placing flour paste cards or trap jars. Reduce harborage by enclosing vulnerable materials in insect-proof containers. Reduce relative humidity and raise or lower temperatures to make environmental conditions unfavorable for silverfish.

If a problem arises, evaluate the magnitude of the problem before initiating intervention tactics.

After other management options have been considered and implemented, if appropriate, select approved chemical controls that are least disruptive to the environment.

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Spiders and Scorpions

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for spiders and scorpions. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Most people are familiar with the general appearance of both spiders and scorpions. Spiders and scorpions are both arachnids, which is a group of animals that also includes mites, ticks, and harvestmen (daddy longlegs). The arachnids are closely related to insects. Both spiders and scorpions, like insects, have a hard external body, but spiders and scorpions have four pairs of legs while insects have three pairs.

Many people may fear spiders and scorpions because of misunderstandings concerning their dangerous nature. In reality there are only a few species of spiders and scorpions that warrant caution. Both spiders and scorpions are a normal and desirable part of the ecosystems in which they occur. They feed on other insects, including species which are pests of plants and nuisance species such as biting flies, as well as other spiders and scorpions. Therefore, spiders and scorpions are generally considered to be beneficial organisms.

Despite the generally benign nature of most scorpions and spiders, bites or stings by some species can be life-threatening to small children, the elderly, or people who are hypersensitive. There are three spider groups of medical importance: the widow spiders (including the black widow, *Latrodectus mactans*), the brown recluse spider, *Loxosceles reclusa* (and the related *Loxosceles laeta*), and the aggressive house spiders (genus *Tegenaria*). There is only one genus of scorpion found in the United States that is of medical importance (*Centruroides*), commonly known as bark scorpions. None of these dangerous species of scorpions and spiders bite or sting humans without provocation. The majority of bites or stings occur because the spider or scorpion has been sat, rolled, or stepped on, or because attempts were made to pick up the spider or scorpion.

In this module, the biology and habits of the major species of spiders and scorpions that are potential health hazards will be discussed in detail. Control measures and education programs to increase public awareness of the potential hazard of these species are outlined. The geographic regions in which specific dangerous species of spiders and scorpions occur are identified and the most common places in which humans are likely to be exposed are discussed. This report **does not** include emergency medical procedures for care of persons bitten or stung by these dangerous species. However, some information is provided on how to determine if medical treatment should be sought. THIS INFORMATION IS PROVIDED ONLY AS A GUIDE. If in doubt, always consult medical professionals.

IDENTIFICATION AND BIOLOGY OF SPIDERS AND SCORPIONS

Scorpions are predaceous on various small invertebrates (insects, arachnids and related arthropods) and vertebrates (small lizards and rodents). Scorpions can withstand long periods of starvation (up to five months) without any noticeable effect. In areas where there are occasional subfreezing temperatures scorpions will hide in warmer rock and bark crevices (Ebeling 1975). Scorpions become inactive at temperatures of 40-50^{o}F (4-10^{o} C) and at extremely high temperatures (Polis 1990).

Scorpions give birth to live young. Immediately following birth, the young scorpions crawl onto the back of their mother where they remain for 5-15 days. The young scorpions are white and soft during this time. Following pigment development (tanning), the young scorpions will leave their mother's back and begin to forage for food. Scorpions may live for several years depending on the availability of food and water.

Geographic Distribution

In the United States, scorpions are most abundant in the arid and semi-arid regions of the Southwest. No scorpion species occur in the Rocky Mountains, North- Central,or Northeastern United States Only one species which occurs throughout most of the Southeastern United States, and one species occurs in northern Florida, while three species occur in southern Florida. Southern Nevada, southeastern California, and central Arizona have the highest diversity of scorpion species in the United States (Polis 1990).

The bark scorpion, *C. exilicauda* (=*sculpturatus*), which is the only American species of scorpion whose sting may be life-threatening, occurs primarily in the southeastern part of Arizona, but may also be found in southern New Mexico and southern California. However, a related but less venomous species, *C. vittatus*, occurs in Texas, especially in Big Bend National Park, and in the southeastern United States from South Carolina south to Florida and west to New Mexico. (*C. vittatus* was formerly divided into three species, *C. vittatus*, *C. partherienois*, and *C. chisosarius*).

Habitat

Scorpions are nocturnal and hide in crevices and shaded areas during daylight hours, in part to reduce loss of moisture. Even during evening hours, scorpions spend most of their time in burrows or hiding places (Polis 1990). Despite occurring in arid regions, scorpions need to drink water in addition to obtaining it from their food. Because of this need for moisture, scorpions may be more common near natural and artificial sources of water at night. During seasons when scorpions are active, most desert scorpion species are most active during the early hours of the evening (prior to midnight) (Polis 1990). During daylight hours, scorpions hide in areas where protection from daytime heat and sun, and water are available. Unfortunately, areas occupied by humans in arid regions are also associated with natural and artificial water sources, increasing the likelihood of human-scorpion encounters.

The bark scorpion is almost always found associated with trees, especially in riparian habitats. This species is commonly found in mesquite, cottonwood, and sycamore groves. It is a climbing species and is almost never found hiding in burrows, except during periods of hibernation. Its climbing habit distinguishes it from all other scorpions in its geographic range (Smith 1982).

Scorpions are attracted to water sources in buildings. Bathroom and kitchen areas are the most frequent places scorpions can be found at night in buildings. During the day, scorpions will seek out hiding places such as cracks and crevices in the floor, cabinets, attics, wall voids, and crawl spaces. Scorpions are most often a problem in buildings in newly developed areas (within three years). This is attributable to the disturbance or destruction of the scorpion's territories (Smith 1982). Additionally, buildings near arroyos or dry riverbeds may experience an influx of scorpions during periods of rain as the scorpions seek out higher ground. Scorpions in buildings are not likely to meet their normal requirements of temperature and prey density (Smith 1982).

MONITORING AND THRESHOLDS FOR SCORPIONS

The presence of scorpions in an area can be detected by trapping and visual scouting. Pitfall traps (a small hole dug into the ground and covered with a board, rock, etc.), although not scorpion specific, may help to identify the presence of scorpions. For best results a trap should be set near water sources and checked during daylight hours while wearing leather gloves. This is when scorpions may be hiding in the trap, except for the bark scorpion which hides under bark on trees during daylight hours. Visual scouting for scorpions can be done during both daylight and dusk (or early evening) hours. During daylight hours a visual search under rocks, loose bark, and other debris (while wearing leather gloves) can confirm the presence of scorpions. At night, the inside of buildings and outside areas may be searched using a ultra-violet fluorescent light fixture. Scorpions glow brightly under black light and are extremely conspicuous even from yards away (Smith 1982).

NON-CHEMICAL CONTROL OF SCORPIONS

Programs to educate the public should be implemented in areas where scorpions are known to occur. These should include identification of scorpions, especially recognition of the dangerous species occurring in the region. People should be encouraged to avoid risky activity in areas where scorpions have been observed. The program should also educate the public about the beneficial role that scorpions play in the ecosystem, and the importance of scorpions as natural enemies of other arachnids and insects. Lastly, preventative behaviors should be outlined and the groups of people most at risk identified.

Additional precautionary methods that should be included in the education program are: 1) wearing leather gloves when moving objects and collecting firewood at campsites or in outdoor areas, 2) when camping, invert and shake out clothes, sleeping bags and other items that have been in contact with the ground or trees, and shake out shoes before putting them on, and 3) always wear shoes when walking at dusk or at night.

The best methods for controlling scorpions are 1) those that alter the habitat where human contact is likely in order to make it less suitable for scorpions and 2) the creation of barriers that restrict the movement of scorpions into buildings and areas where contact is likely. Cultural methods such as sanitation and elimination of harborages have been found to be effective in reducing scorpion numbers. Barriers to movement of scorpions into dwellings can also be effective in reducing exposure to scorpions. Barriers for scorpion exclusion include caulking windows and holes around plumbing.

Cultural Control

Sanitation and removal of debris are the primary methods recommended for the control of scorpions. Firewood should be stored away from the sides of buildings and off the ground. Other debris such as loose boards, rock piles, and trash should also be moved away from buildings. Shrubs should be pruned so that they do not make contact with the exterior of buildings.

Elimination of sources of open water may also reduce the occurrence of scorpions. Proper maintenance of toilets and plumbing to reduce leaks and coating the inside lip of toilets with petroleum jelly will reduce access of scorpions to water. Drains should be screened or plugged when not in use to prevent access from the outside.

In order to create barriers to scorpion movement into dwellings, window frames and screens should be periodically checked for holes large enough for scorpions to enter through. Screens should be repaired and window frames caulked to fill all gaps. Baby cribs and cots can be protected by placing the legs into clean widemouth jars (scorpions cannot climb clean glass surfaces). Holes associated with wiring and plumbing should also be caulked to fill gaps.

CHEMICAL CONTROL OF SCORPIONS

There is little evidence that chemical control tactics are effective against scorpions so they should only be considered as a last resort. However, chemical control methods have been used to control scorpions in areas in which infestations are already identified. There are chemicals registered for the control of scorpions, but proper application is essential for adequate control.

Application of insecticides during daylight hours is largely ineffective against scorpions since scorpions are only active at dusk and at night. Some residual insecticides are registered for use; these may be the method of choice if scorpions are a persistent problem inside buildings (after attempts have been made to exclude them through cultural methods). If scorpions are found in buildings or are frequently found in outside areas where visitors are likely to be active after dark, the use of pesticides may be necessary. If pesticide use is considered, they should be applied to all potential hiding places and points of entry (including, but not exclusively, wall voids, cracks and crevices, attics, and window sills). Dusts are preferred because they can be blown into wall voids, etc.

Consult your regional Integrated Pest Management coordinator to determine which pesticide, if any, is best suited to your integrated pest management program.

IDENTIFICATION AND BIOLOGY OF SPIDERS

The Brown Recluse Spider

The brown recluse spider, *Loxosceles reclusa*, and a related species, *L. laeta,* are also commonly referred to as violin or fiddleback spiders because they have a dark fiddle shaped pattern on their upper body. They vary in color from tan to dark brown. A second identifying characteristic of the brown recluse spider is the presence of only three pairs of eyes. (Most spiders have eight.) (Akre and Catts 1990.)

The brown recluse spider is sedentary and builds an irregular web that is often not recognized as a spider web. Females lay eggs in flattened egg sacs that are frequently attached to the underside of objects. Mating in this species occurs from February to September. Up to 40 spiderlings may hatch from a single egg sac. A single female may produce up to five egg sacs in a summer. Females can live up to four years, males less.

Indoors, the brown recluse can usually be found in infrequently disturbed areas away from light sources, such as behind pictures, beneath or behind furniture, in boxes, in clothing, among stored papers, between the corrugation of boxes, and under food sacks (Hite et al. 1966).

The natural habitat of the brown recluse includes the underside of rocks, loose bark, and crevices in decaying logs (Hite et al. 1966). However, many outdoor refugia provided by the activities of man are frequently inhabited by the brown recluse spider. For example, a survey of piles of junk in Kansas, piles of old tires and inner tubes, furniture, old boards, and trash were found to be inhabited by the brown recluse. Once the debris was removed and the natural vegetation returned to the area, the colony was eliminated.

The brown recluse spider occurs in a region roughly delineated to the north by the northern boundary of Illinois, to the west by the western boundary of Kansas and Oklahoma, and to the east by Tennessee (Akre and Catts 1990). Additionally, single specimens of the brown recluse, presumably artificially transported to these areas, have been reported from Washington D.C., Arizona, California, Florida, New Jersey, North Carolina, Pennsylvania, and Wyoming (Gladney 1972). *L. laeta* , native to South America, has also been reported in the United States in Massachusetts (Gertsch 1967) and California (Keh 1970). Both of these reports of *L. laeta* indicate that it has been transported into the United States by people. It is unlikely that *L. laeta* has become established in the United States, but it may be an occasional problem in areas where products are frequently shipped from South America.

The Widow Spiders

There are five species of widow spiders (*Latrodectus*) in the United States. The combined geographic range of these spiders encompasses the entire United States. Three of these species can generally be considered to be "black widows." Females of all of these species are metallic black with reddish marks commonly forming an hourglass shape on the underside of their thorax. The most well-known species, the common black widow spider, *Latrodectus mactans* occurs from southern New England to the southern United States The Northern widow, *L. variolus* , occurs from the mid- Atlantic states north to Canada. The western widow, *L. hesperus*, occurs west of the Rocky Mountains. Two additional species, the brown widow, *L. geometricus*, and the red widow, *L. bishopi*, are tropical species whose United States distribution is restricted to southern Florida. *Latrodectus geometricus* is another introduced species that primarily occurs in domestic situations, but its distribution is sporadic (Gertsch 1979).

Widow spiders are cobweb builders; a typical web of a widow spider is a small, tangled maze of coarse fibers that are made in dark corners or crevices. Frequently these webs are made near ground level. These webs may not even be recognizable as an active spider web. Eggs of the widow spiders are laid in sacs of silk within the female's web. A single egg sac may contain up to 400 eggs. The eggs of widow spiders hatch in three to four weeks. The hatchlings are highly cannibalistic and therefore most of the young will be consumed by their brothers and sisters. Web- spinning spiders such as the widow spiders are not active outside of their webs. This is especially true of the western widow spider which creates webs primarily in cracks and crevices.

Aggressive House Spiders

The aggressive house spiders are in the genus *Tegenaria.* Only one native species, *Tegenaria chiricahuae*, occurs in the United States, but at least six introduced species of *Tegenaria* now occur in the United States. These spiders as a group are often referred to as funnel-web spiders. They build funnel shaped webs in dark, moist areas such as basements and crawl spaces, and sit in these webs and wait for prey to walk by. Generally, these spiders are yellow to pale tan in color with long legs. These spiders occur in highest frequency in July through September and reproduce during this period. Females produce an egg sac that is placed near the opening of the funnel in their webs. Eggs hatch the following spring.

Although the bite of these species is not considered to be as dangerous as that of either the brown recluse or widow spiders, it can cause a similar ulceration of the skin as the brown recluse and may involve systemic reactions. The species that cause the worst bite reactions are found in the northwestern United States; *Tegenaria agrestis* occurs from Idaho to Vancouver and Winnipeg in Canada. It builds a web at or near ground level, and rarely climbs up vertical surfaces (Akre and Catts 1990). This spider is called an aggressive house spider because it will bite with little provocation if cornered or threatened. This may be related to their hunting strategy and may increase the likelihood that humans will be bitten by these spiders.

NON-CHEMICAL CONTROL OF SPIDERS

Sanitation and habitat modification are key tactics for control of spiders indoors. This includes vacuuming in corners, window sills, and attic areas, and keeping premises free of unneeded, unwanted items such as undisturbed clothing, papers, and other litter. Indoor habitat modification that creates a barrier to the movement of spiders into buildings is also a key tactic to effective spider control.

Cultural Control

Sanitation is one of the key methods of controlling spiders in buildings. The corners and crawl spaces of buildings should be kept free of spider webs. This may be accomplished by simply dusting these areas or by using a vacuum to remove existing webs. Vacuuming removes active spider webs, adult spiders, and spider egg sacs. Living spiders will desiccate quickly in the vacuum bag, but depending on the design of the vacuum, it may be useful to empty the bag immediately after use in order to prevent the spiders' escape (Akre and Catts 1990). Removing litter such as newspaper and wood from the interior and the sides of buildings is also crucial for effective elimination of spiders. In addition to sanitation, creating a physical barrier to movement of spiders into buildings is also an effective management technique. Pruning shrubbery and other plants away from buildings will also limit the access of spiders to buildings.

Barriers also limit access of buildings to spiders. Caulking, repairing screens, and filling cracks and crevices around windows, doors, and foundations with materials such as expanding polyurethane foam will exclude many spiders from buildings. Common areas to inspect for holes and gaps include entry holes for plumbing and electrical lines, and window and door casings. Window and door screens should be repaired to fill in holes large enough for entry of spiders. Gaps in the wall boards and ceiling-wall interfaces should be closed, and door and window casings should be filled with caulking or a foam insulation material. Foam insulation material can also be used to fill wall voids and crawl spaces if spiders come in through these areas. Spiders can easily gain access to buildings through gaps beneath doors. Placing a piece of weather stripping under a door so that there is no gap between the bottom of the door and the floor when the door is closed will alleviate this problem.

If crawl spaces are a breeding area for spiders, the reason is usually excess moisture. By eliminating moisture from crawl spaces, spiders can be eliminated. Placing plastic over bare soil can eliminate moisture in some areas, such as beneath cabins. The key to many moisture problems is to increase venting. Therefore, opening up ducts under a foundation may eliminate moisture from a crawl space, without allowing increased access of the building for spiders.

Additional precautionary measures which may reduce the risk of being bitten by spiders include wearing shoes at all times, using leather gloves when moving rocks, wood or other debris, and shaking out sleeping bags and clothing before using them.

CHEMICAL CONTROL OF SPIDERS

Chemical control of spiders inside of buildings is not recommended and should be considered only as a last resort. Residual sprays are not recommended for use in buildings that are occupied, or are to be occupied in the near future. If residual materials are used in buildings not currently occupied or in areas where other methods fail, applications are recommended only along baseboards, door casements, and corners, and only where spiders are present (Akre and Catts 1990).

Chemical control using a long-lasting residual pesticide can be effective in controlling populations of spiders outdoors. Many residual materials are registered for control of spiders. Consult your regional Integrated Pest Management coordinator to determine which registered material is best for your integrated pest management program. Problem outdoor areas usually needing treatment include porches, garages, eaves of the roof, crawl spaces, and other areas beneath buildings (Bennett and Williams 1989).

First Aid

For any bite or sting it is important to reduce stress and help the inflicted person to relax. There is evidence that this will reduce the toxic effects of some bites and stings (Ebeling 1975). An ice cube may be applied for a short time to reduce the pain at the site of the bite or sting; this does not reduce the effect of the bite, but may make the afflicted person more comfortable. (DO NOT IMMERSE THE WHOLE LIMB IN WATER.) If in doubt about the seriousness of a bite or sting, or if a person is bitten or stung by any of the medically important species discussed in this module, contact your local poison control center or a physician immediately. Also, collect the scorpion or spider in question if possible to assist in the treatment of the sting. For further discussion of medical treatment and the progress of envenomation by scorpions, or spiders, see Smith 1982, Ebeling 1975, or Polis 1990 for treatment of scorpion envenomation.

The sting of most scorpion species and the bite of most spider species are not considered to be dangerous. However, if a person is stung by a scorpion in an area in which the bark scorpion occurs medical attention should always be sought since its sting may be life-threatening (Smith 1982). Additionally, any bite or sting may elicit an unusual allergic reaction by persons who are hypersensitive to the bite of a specific species. For this reason all bites must be examined to ensure the safety of those involved. A hyperallergic reaction can lead to anaphylactic shock and in very severe cases, respiratory distress may develop. It is not unusual for a person to have some pain and numbness in the same region as the site of the bite. However, if, for instance, a person is bitten on their hand and their legs begin to swell, this is indicative of a systemic reaction, and this person should receive medical attention as soon as possible. People who are known to be hypersensitive to other stinging insects such as bees and wasps are not necessarily hypersensitive to spider bites or scorpion stings. Likewise, each spider or scorpion has a very specific type of venom and a person may be sensitive to the venom of one species and not sensitive to the venom of a closely- related species. Lastly, some anti-venoms are available for treatment of some bites and stings, but their availability is variable. Contact your local poison control center for information regarding anti-venoms if dangerous spiders or scorpions are a problem in your region.

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Thistles

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for thistles. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Thistles include many species of composite plants in the subfamily Cynareae. Although most species in this subfamily are native and are beneficial for wildlife, a number of introduced species are well known as serious weeds of crops and rangelands. The pest status of introduced species results from the lack of population suppression exerted by natural enemies (e.g., insect herbivores and diseases). Thus, long-term goals in thistle management emphasize biological and cultural controls, although emergency intervention with chemical or mechanical controls may be necessary.

IDENTIFICATION AND BIOLOGY OF THISTLES

Five species of thistle are currently considered major pest weed species by the National Park Service and are under chemical, biological, or cultural control programs in North America. Other species, such as *Carduus acanthiodes* (plumeless thistle), *C. crispus* (welted thistle), and *C. macrocephalus* (nodding thistle), may be pest weeds in local areas. The five species listed below were introduced from Europe and North Africa into North America.

Musk thistle (*Carduus thoermeri* Weinmann). Originally thought to be *C. nutans* (L), *C. thoermeri* is only found in small isolated pockets. The musk thistles are a complex of several species or subspecies, the taxonomy of which is unclear. See McCarty and Lamp (1982) for details. Musk thistles are found throughout the United States, and are especially common in Southern California and Midwestern and Appalachian regions. Musk thistles are biennial thistles that flower in May-June.

Italian thistle. (*Carduus pycnocephalus* [L]). Italian thistle is found in California (mainly in coastal counties) and are rare in other areas of the United States. Italian thistles are summer annuals or may be biennials in dry habitats.

Canada thistle. (*Cirsium arvense* [L]). Canada thistle is found throughout North America except in Alaska, and is most common in northern tier of states and southern Canada. Canada thistles are perennial thistles which flower in June-October.

Bull thistle. (*Cirsium vulgare* (Savi) Tenore). Bull thistle is found in southern Canada and throughout the United States. Bull thistles are biennial thistles which flower in July-September.

Milk thistle. (*Silybum marianum* [L]). Milk thistle is found in California, mainly in the coastal counties and drier areas. Milk thistles are winter annuals or may be biennials in dry habitats.

See Fernald (1950), pages 1538-1542, and Peterson and McKenny (1968), pages 302-306, for descriptive keys and illustrations of thistle species. See also Moore and Frankton (1974) for detailed keys to species.

Thistles are pioneer species and are most often found in sites where the ground cover has been disturbed by grazing, erosion, traffic, or other means. Thistles reduce the use of an area for grazing or recreational purposes because of the prominent spines on leaves, stalks, and blooms. Livestock do not eat thistles and will not graze between thistle plants on more desirable forage (Batra 1982).

Each thistle produces many seeds, often in excess of 10,000 seeds per plant. The fine filaments or pappus (thistle down) of the seed coat permit windborne dispersal over long distances to suitable habitats. Reinfestation occurs from roadsides or other areas where control is not practiced or by long-lived seeds stored in the soil from previous years. Newly germinated thistle seeds require considerable light and usually become established on disturbed areas of pastures or croplands where competition is limited during the seedling stage (Hodgson 1968). Foliar growth occurs during the spring, summer, and fall. The amount of growth and rate of new establishment varies from region to region according to the geographic, ecologic, and climatic characteristics of each region.

Losses in cultivated crops are as high as 60% at usual levels of infestation (25 shoots of Canada thistle per square yard). Losses in productivity of forage grasses from Canada thistle at a density of less than two shoots per square meter are as high as 15% (Hodgson 1968).

The introduced thistles represent a range of life histories, and the life history of each species may vary depending on habitat conditions:

Summer annuals grow each spring or summer from seed. They grow, mature, produce seeds, and die in one growing season. Seeds generally overwinter before germinating the following spring.

Winter annuals germinate in late summer or fall from seed, then mature and produce seed the following spring or summer. Seeds are dormant during the spring.

Biennials germinate any time during the growing season. They usually produce a rosette of leaves close to the soil during the first season, then flower (using energy and nutrients stored during the first season's growth), mature, and die during the next year.

Perennials become established by seed or vegetative parts (e.g., roots, tubers, or rhizomes). Once established, they live for more than 2 years, and often for many years.

MONITORING AND THRESHOLDS FOR THISTLES

Thistles are relatively conspicuous weeds and in most cases periodic visual inspections should be sufficient to monitor thistle populations. The permanent plot technique is a good way to monitor thistle populations after they have become established and while they are being controlled. A representative section of the field is marked off and thistles are counted and mapped and notes made on their condition (height, flowering, etc.). Monitor on a regular basis (weekly, biweekly, monthly). Keep careful records, note when treatments take place, or when biological controls are introduced (naturally or artificially). Study of records, over time, will show population trends and indicate whether or not control strategies are successful.

A variation on the above is the use of photo plots. Take a series of photographs of the sample plot showing the density of thistles and condition. Include in the photo an object of known size (person or measuring stick) to indicate thistle size. Also include in the frame a sheet of paper with the date in bold letters. All photos should be taken from the same location with the camera pointed in the same direction and with lenses of equal coverage. This method is especially useful in monitoring the effects of control measures over the course of several seasons.

Many states have laws requiring the control or removal of pest thistle species before they flower, whenever and wherever they occur. In these states, and in most other instances, the threshold action level is one or more weed thistles.

NON-CHEMICAL CONTROL OF THISTLES

The primary control strategy for annuals and biennials is seed management while the control strategy for perennials must include depletion of plant reserves. Long-term strategies for thistle control depend on biological and cultural controls. Generally, no one technique will provide adequate control. Currently available biological controls using insects require several years for establishment of the insect, and even longer for control. Most successful programs combine biological control with cultural controls such as timely mowing or reseeding with competitive desirable plants. Suppression of thistles may require altering land use.

Biological Control

Pest species of thistles have been introduced into North America without their compliment of natural enemies. In Europe, *Carduus* thistles are attacked by approximately 340 species of insects and 7 fungal pathogens. Current research in biological control is an attempt to reunite natural enemy species with their hosts. Biological control agents seldom eliminate pest thistles from an area, but can reduce populations below set economic thresholds.

Imported thistles have been the subject of biological control programs for several years. The following is a brief description of several biological control agents.

Rhinocyllus conicus, a European weevil that feeds on developing seed heads has been introduced into the United States and Canada for control of *Carduus* thistles, particularly the musk thistle. It has also been introduced for control of Italian and milk thistles in the western United States. In the absence of *Carduus* thistles, *R. conicus* will feed on Canada and bull thistles, but control is not as complete as on its primary host. *R. conicus* deposits eggs on bracts and flower stems. Larvae feed beneath developing seeds, destroying them. Pupation occurs in the flowers, and adults emerge in mid-summer. Adults hibernate in overwintering floral rosettes. There is one generation per year. Release of *R. conicus* on National Park Service lands is not being allowed at this time due to possible impacts on native plants.

Trichosirocalus horridus, another European weevil, has also been introduced for control of *Carduus* thistles. This insect feeds primarily on the root crowns of musk and Italian thistles. *T. horridus* has been released in Canada and most of the United States. It has not been introduced on the west coast of the United States because it has been shown experimentally to feed on artichoke. However, it is not considered to be a pest in artichoke-producing areas of Europe, and further studies are being carried out. *T. horridus* deposits eggs on leaf ribs and larvae migrate to the root crown where they feed. Pupation occurs in the soil. Adults feed after emergence and overwinter in the rosettes. Weevils from populations in southern Europe and from central Europe have been introduced into the United States. Southern European weevils mate in autumn, oviposit from mid-December to March, and adults emerge in April and June. Central European weevils mate in spring and oviposit in May to June. Adults emerge in September and hibernate until the spring thaw. These two populations are currently undergoing further study to develop more effective control for thistles. Stoyer and Kok (1987) suggest that a combination of *T. horridus* and sublethal dosages of 2,4-D herbicide could aid in *Carduus* thistle control.

For control of Canada thistle, *Altica carduorum*, a European weevil has been imported into North America. Adults feed throughout the summer on leaves, defoliating the plant and weakening it. Although Canada thistle is seldom killed outright by this weevil, the continued stress upon it reduces the number and vigor of vegetative shoots and reduces seed production. Although repeatedly released in North America, this species is not yet well established (Batra et al. 1981).

A second weevil, *Ceuthorhynchus litura*, that feeds on leaves and root crowns of Canada thistle is established and providing some control in Canada, Idaho, Montana, and California (Rees 1990).

A tephritid fly, *Erophora cardui*, that feeds on Canada and bull thistles was released in 1973 and is established in British Columbia.

Cassida rubiginosa, a chrysomelid beetle that feeds on leaves of *Carduus* and *Cirsium* thistles, has been established in North America since 1927 (Batra et al. 1981).

Several other species of insects, mostly seed-head weevils, are currently being studied for possible importation and release for biological control of thistles in the United States.

Two fungal pathogens that are spread by thistle feeding insects are also being considered for release in the United States. Rust fungi in the genus *Puccinia*, which attack the leaves of the basal rosette and underground basal parts, have been introduced into Canada. Further studies are required to determine their effectiveness.

Ustilago cardui, a smut fungus, has been observed to attack late maturing seed heads of *Carduus* thistles in Europe. Seed production is stopped in infected plants, giving full control. This fungus compliments control by *R. conicus* , which feeds on early flower heads (Bolt 1978).

Consult your National Park Service regional Integrated Pest Management coordinator for further information on biological control for thistles in your area.

Cultural Control

In areas that are grazed, eroded, or subject to heavy traffic, the grass cover may not be dense enough to prevent establishment of thistles. Rotational or deferred grazing, water conservation, erosion control, redirection of traffic, and sound pasture and turf management practices can reestablish heavy grass cover and prevent thistle establishment (Bendall 1973, Trumble and Kok 1982, Kok et al. 1986).

Mechanical Control

Cutting or removing thistles (where feasible) can be effective in reducing thistle populations. Annual and biennial thistles, if mowed within two days of flowering of the terminal blooms, will not produce seed or regenerate significantly. Timing in mowing is important; if mowing occurs four days after terminal bloom anthesis (full flowering), significant amounts of seed are produced. Since thistle stands mature at different times, careful monitoring and proper timing are necessary for mowing to be a viable option in an Integrated Pest Management program. However, even if mowing is done late and seed is produced, mowing the stalks will reduce seed dispersal and seed production, keeping infestations from spreading widely (McCarty and Hattling 1975).

Canada thistle, a perennial, is difficult to control by mechanical methods. Occasional cultivation may increase sprouting from broken roots due to its ability to propagate vegetatively. However, repeated cultivation can significantly reduce infestations if begun when plant reserves are at their lowest stage in early spring (early bud stage), before the shoot leaves can furnish energy to the roots in amounts greater than the roots require for production of new growth. Cultivation should start in early spring by plowing and disking. When new shoots appear, the area should be cultivated 3"to 4" inches deep every 20-21 days to destroy new shoots. Up to 90% or more of a Canada thistle infestation can be eliminated in a single season of cultivation when properly performed. Remaining plants can be eliminated by continuing cultivation in the following spring (Hodgson 1968). Hodgson (1968) reports excellent control of Canada Thistle in alfalfa fields mowed for hay twice a year.

Mechanical controls are compatible with biological controls if the mechanical controls are used early in the season to stress the plants, and natural enemies are allowed to enter the system to further weaken and eliminate thistles. Mechanical controls combined with chemicals may be successful in some cases. In most cases, however, combining a chemical and biological control is a more viable approach to thistle management.

Controlled burning may only damage the above ground portion of the thistle allowing rapid regrowth from the root section or from seed. Fire should be used only in combination with other control measures.

CHEMICAL CONTROL OF THISTLES

Several herbicides are useful for thistle control and your regional Integrated Pest Management coordinator should be consulted for more information on these. Spot treatments, rather than broadcast treatments, are preferred. Chemical control for annuals, biennials, and perennials must be initiated before the plants blossom and produce seeds. Young plants are most susceptible to control with chemicals. Best results are obtained when plants are in their initial and heaviest growth stage. The use of herbicides provides a quick and easy (albeit expensive under largescale operations) method of control, but without a long-term strategy herbicides often lead to greater problems because of their effect on other plant species, the development of resistance, and the lack of susceptibility of certain life stages of thistles.

Trials combining herbicides (usually 2,4-D), and biological control agents (*R. conicus* and *T. horridus*) have shown the two to be compatible if precautions are taken (Trumble and Kok 1980). Field and laboratory tests have shown that spring application of 2,4-D (when blooms are beginning) provides the most effective thistle control, and causes the fewest adverse effects on thistle weevils; *R. conicus* adults and *T. horridus* pupae, the only life stages likely to be exposed to such spraying, are relatively unaffected by the herbicides. Adults of both species will move to unsprayed plants, thus increasing biological control in nearby areas where herbicide treatment is not feasible or economical. Tests to determine compatibility of biological control agents with herbicides other than 2,4-D are still in the planning stage.

SUMMARY

To summarize, the following steps are recommended to manage thistles:

1. Monitor infestations over time with the use of maps, plots, or photographs.

2. The primary control strategy for annuals and biennials is seed management, while the strategy for perennials must include depletion of plant reserves.

3. Use biological controls in your area if possible. Check with your National Park Service regional Integrated Pest Management coordinator for details.

4. Use cultural controls to reestablish dense grass or ground cover in order to prevent or reduce

thistle establishment.

5. Cut, mow, or otherwise remove thistles, if feasible. Thistles should be cut before the flowering of terminal blooms to prevent seed production.

6. Use appropriate herbicides on a spot treatment basis. Time applications to control thistles at prebloom stage and for compatibility with natural enemies.

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Ticks

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for ticks. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Ticks are external parasites on mammals, birds, reptiles, and amphibians. Both males and females feed on blood.This module describes the biology and management of five species of ticks commonly found in park settings. These ticks are all species which vector a disease, are capable of transmitting a pathogen to humans, or may in some other way affect human health. They are the Lone Star tick, *Amblyomma americanum* (L), American dog tick, *Dermacentor variabilis* (Say), Rocky Mountain wood tick, *Dermacentor andersoni* (Stiles), deer tick, *Ixodes dammini* (Spielman, Clifford, Piesman, and Corwin), and *Ornithodoros* spp. For each species of tick, the geographic distribution, habitat, hosts, life cycle, seasonal abundance, responses to environmental factors, and direct and indirect medical effects are described. Information concerning the removal of ticks, outbreaks of tick-borne diseases, and natural enemies are presented. Tick management approaches including methods of population monitoring, decision-making, and intervention are described. All of these tick species are attracted to carbon dioxide and generally prefer low light intensity, high relative humidity, and protection from constant breezes. Temperature and humidity are the two most important environmental factors affecting survival.

IDENTIFICATION AND BIOLOGY OF TICKS

The identification of medically important species of ticks can be done by local diagnostic facilities at universities or state agencies or with the aid of publications such as Keirans and Litwak (1989), Sonenshine (1979), and the United States Department of Health, Education and Welfare (1967), which provide keys and descriptions. *Ixodes dammini* was first described in 1979 and will appear as *Ixodes scapularis* in works prior to this date. A concise review of tick biology, management, and medical importance was provided by Goddard (1989).

Ticks are considered harmful because they transmit diseases. Like many other organisms, however, their role in the food chain serves a positive ecological function. Ticks are an essential food source for many reptiles, birds, and amphibians.

Lone Star Tick

This tick species occurs from central Texas east to the Atlantic coast and north to Iowa and New York; it has also been reported in northern Mexico. The Lone Star tick is found in wooded areas, especially where there is dense underbrush, but
it is also found in scrub, meadow margins, hedge rows, cane breaks, and marginal vegetation along rivers and streams. The immatures and adults feed on a wide variety of mammals (including humans) and ground-feeding birds.

Each female produces 3,000-8,000 eggs, which are deposited under leaf and soil litter in middle to late spring. Incubation may take 30 days or longer, depending on temperature. The newly hatched six-legged immatures, also known as larvae or seed ticks, feed for 3 to 7 days on a host. After full engorgement the larvae drop from the host into vegetation and shed their skins 9-27 days later. The eight-legged immatures that emerge are called nymphs. These attach to a second host and feed for up to 38 days; the nymphs then detach and rest for 13-46 days before they shed their skins to become adults. Adults attach to a third host, feed for 6-24 days, and detach. Oviposition occurs 7-16 days after the last blood meal. Larvae may survive for 2-9 months, and nymphs and adults for 4-15 months each (Goddard 1989); the life cycle may take up to 2 years to complete. Lone Star tick nymphs can move very quickly and may cover a person's legs or arms in less than five minutes. This is a good behavioral characteristic to note to aid in identification of this tick species.

Adults and nymphs are active from early spring through midsummer, while larvae are active mainly from late summer to early fall. Low humidities and high daytime temperatures restrict the occurrence and activity of these ticks (Goddard 1989).

Lone Star ticks transmit Tularemia to humans. Lone Star ticks infected with the agents of Rocky Mountain spotted fever and Lyme disease occur in nature, but the species does not appear to be epidemiologically important in the transmission of these diseases (see Goddard 1989).

American Dog Tick

The American dog tick is found throughout the United States except in parts of the Rocky Mountain region. It also occurs in parts of Canada and Mexico. Its habitat includes wooded areas, abandoned fields, medium height grasses and shrubs between wetlands and woods, and sunny or open areas around woods. Larvae and nymphs feed primarily on small mammals (especially rodents), while the adults feed mainly on dogs, but will readily bite humans.

The female lays 4,000-6,500 ellipsoidal eggs over a 14-32 day period and then dies. The eggs usually hatch in 36-57 days. Larvae usually engorge for 3-5 days, nymphs for 3-11 days, and adult females for 5-13 days. Unfed larvae can live up to 15 months, nymphs 20 months, and adults 30 months or longer. Mating takes place on the host (Goddard 1989, Metcalf and Flint 1962). Adults are active from mid-April to early September. Nymphs are active from June to early September and larvae from late March through July. High light intensity and low relative humidity stimulate questing behavior (Newhouse 1983).

This species is the primary vector of Rocky Mountain spotted fever in the eastern United States, and can also transmit Tularemia and cause tick paralysis.

Rocky Mountain Wood Tick

This tick is found from the western counties of Nebraska and the Black Hills of South Dakota to the Cascade and Sierra Nevada Mountains, and from northern Arizona and northern New Mexico in the United States to British Columbia, Alberta, and Saskatchewan in Canada. Their habitat is primarily fields and forested areas. This species is especially prevalent where there is brushy vegetation that encourages the small mammal hosts of immature ticks and sufficient forage to attract the large hosts of the adults. Immatures feed mainly on small mammals such as ground squirrels and chipmunks, and adults on cattle, sheep, deer, humans, and other large mammals.

Females lay about 4,000 eggs in plant debris on the soil or in crevices in construction materials, usually in masses of hundreds at a single location. Unfed larvae may live for 1-4 months, nymphs for 10 months, and adults for more than 12 months (Goddard 1989). Adults and nymphs can be found from March to mid- summer. Larvae are active throughout the summer and are associated with cool soil temperatures, shallow soil, abundant leaf litter, and high relative humidity.

This species is the primary vector of Rocky Mountain spotted fever in the Rocky Mountain states and is also known to transmit Colorado tick fever and Tularemia. It also carries tick paralysis in the United States and Canada.

Deer Tick

The deer tick is found in eastern North America including the New England, mid-Atlantic, and southeastern states, and the midwestern states of Minnesota and Wisconsin. It has also been observed in Michigan, Iowa, Illinois, and Indiana. Deer ticks prefer heavily-forested or dense brushy areas and edge vegetation, but not open areas. An exception to this occurs in upstate New York where the species is found on well-maintained lawns in residential areas. Larvae and nymphs feed primarily on small mammals (especially the white-footed mouse, other rodents, and insectivores), and also on birds, dogs, deer, and humans. Nymphs aggressively bite humans. Adults feed primarily on deer, but also attach to large mammals (foxes, raccoons, opossums, dogs) and humans.

Females lay up to 3000 eggs in soil and litter. Eggs take about 1 month to hatch. Larvae engorge for 2-3 days during the summer, detach, overwinter on the ground, and molt the following spring. Nymphs feed for 3-4 days, detach, and molt in early fall. Adult females engorge for 7-21 days, detach, oviposit the following spring, and die. The life cycle may range from 2-4 years and is regulated by host abundance and physiological mechanisms. Larvae are active from July through September, nymphs from May through August, and adults in the fall, winter, and early spring (October-May).

Distribution is associated with high humidity and mild mean winter temperatures. However, it is not restricted by winter temperatures as areas of tick activity occur in Minnesota and Wisconsin. The requirement for high humidity restricts this tick from spreading to arid areas and high mountains where desiccation is a limiting factor (Lane et al. 1991).

The deer tick is the major vector of Lyme disease in the northeastern and midwestern United States. It is incriminated as the vector of human babesiosis in the northeastern United States.

***Ornithodorus* spp.**

These ticks are the vector of relapsing fever, which has created serious health problems at the Grand Canyon. The relapsing-fever tick, *Ornithodorus hermsi*, is sand-colored before feeding, but turns grayish-blue after it feeds. The adult female is about 1/4" long.

MEDICAL EFFECTS OF TICKS

Ticks may cause paralysis in humans that is reversible when the ticks are removed. Symptoms include paralysis of the arms and legs, followed by a general paralysis which can be fatal if not reversed. The victim may recover completely within a few hours of the removal of the tick. The paralysis may be caused by a salivary toxin transmitted to humans when a tick feeds. Tick paralysis is frequently associated with the attachment of the tick at the base of the victim's skull; however, the illness occurs from attachment to other parts of the body as well. The highest incidence of tick paralysis in north America occurs near the border of British Columbia, Canada, and the northwestern United States.

The two most important tick-borne diseases in the United States are Lyme disease and Rocky Mountain spotted fever. The onset of Lyme disease is usually characterized by the development of a large, red rash which may develop a characteristic clear central area ("bulls eye"), one to two weeks after a tick bite, often in the area around the puncture. Other symptoms include joint pains, flu-like symptoms, and neurological or cardiac problems. The most characteristic symptom of Rocky Mountain spotted fever is a rash on the ankles, wrists, and forehead one to two weeks after the victim is bitten. The rash spreads to the trunk and is accompanied by fever, chills, and prostration. Both Lyme disease and Rocky Mountain spotted fever are transmitted after the tick feeds for several hours. Prompt removal of attached ticks greatly reduces the chances of infection. Both diseases are usually successfully treated with antibiotics in their initial stages. Therefore, early diagnosis is imperative. For this reason, it is recommended that the date of a tick bite be marked on a calendar. If unexplained disease symptoms occur within two to three weeks, a physician should be consulted.

The best means to prevent the transmission of tick-borne diseases and the development of tick paralysis is the **prompt removal of ticks**. This requires regular inspection of clothing and exposed skin for attached or unattached ticks. To remove a tick, grasp it crosswise with narrow tweezers (do not rupture the tick) as close to the point of attachment as possible. Retract or pull tick firmly in the direction of attachment; some back-and-forth wiggling may be necessary. Do not twist or rotate the tick. Do not handle ticks with bare hands because infectious agents may enter through mucous membranes or breaks in the skin. Removed ticks should be immersed in alcohol to kill them. Disinfect the bite site and wash hands thoroughly with soap and water.

The diseases listed above can be fatal. Any case of such a disease should be reported to medical authorities immediately. Frequent or multiple reports of tick-borne diseases should be reported to a National Park Service public health service representative. The representative can recommend actions to control disease outbreaks. Closing affected park areas may be advisable during such periods.

Another important tick-borne disease is endemic relapsing fever. This disease is limited to the western states and is caused by a spirochaete carried by certain ticks in the genus *Ornithodorus*. These ticks are found on tree squirrels (*Sciurus* spp.) and western chipmunks (*Eutamias* spp.). The disease can also be transmitted directly to the tick's offspring. These ticks usually live three to five years. Park personnel and visitors are at increased risk of contracting endemic relapsing fever when they sleep in dwellings that have become inhabited with infected squirrels or chipmunks. As with sylvatic plague, the rodents vacate the building or are killed by the humans who use the buildings. The ticks which remain behind feed on the people using the buildings. Implementation of exclusion efforts will reduce the incidence of ticks.

MONITORING AND THRESHOLDS FOR TICKS

Periodic surveys of potential or known tick habitats can reveal the presence of low-level tick infestations. This permits the application of management procedures to prevent or retard further population increase. Monitoring techniques that have proven effective (Gladney 1978) are as follows.

Examination of personnel for attached ticks. A volunteer wearing protective clothing walks through each sample site and is then inspected. Ticks attached to or walking on the collector's clothing and skin are collected in 70% ethanol for later identification and counting. Careful inspection is necessary to prevent the attachment of unnoticed ticks and possible disease transmission to the collector. Collections can be standardized in relation to time, distance, or area units covered during sampling.

Dragging/flagging. Done by dragging a white cloth over relatively open ground or "flagging" low-level vegetation (i.e., moving the cloth in a waving motion over and through vegetation) in densely brushy ground. Ticks that are questing for passing hosts cling to the cloth and can be removed for identification and counting. The "drag" consists of a 1 yd^2 piece of white crib bedding or corduroy material hemmed on all edges, weighted at one end, and attached to a wooden pole at the opposite end. A rope attached to the two ends of the pole allows the device to be dragged along the ground. Alternatively, the pole can be gripped at one end so that the cloth hangs vertically downwards, and the device used to flag vegetation. Dragging or flagging success depends upon the degree of contact between the cloth and ground or vegetation surface. Useful drag techniques are described by Gladney (1978). The selection of sampling sites may have significant effects on the success of the sampling effort. Sampling sites should reflect favored tick habitats for best success. Sampling should be done under conditions that favor tick presence and activity (e.g., when vegetation is not wet and when ambient temperature is above 50^0 F).

Dry-ice traps. This has been proven to be the most efficient method of tick collection. It is non-destructive to host animals, does not require a human as an "attractant", and gives more reproducible results than dragging. However, the traps need to be kept in the field for several hours (preferably overnight) for best results. Dry ice is available at ice cream and beverage stores. The basic principle is to use carbon dioxide vaporizing from the dry ice to attract ticks onto a white cloth panel on which they are easily visible and can be removed periodically (if the traps are set out for a limited time under periodic monitoring), or onto a platform lined with double- sided sticky tape on which they get trapped (if the traps are set out overnight). Information on trap designs can be obtained from Garcia (1965), Gladney (1978), and Mount and Dunn (1983).

Trapping small animal hosts. Small mammals such as rodents and insectivores can be live-trapped at selected sampling sites, with traps set out in grids or line transects. Trapped animals are anesthetized and searched thoroughly for attached ticks, which are removed using fine forceps. Removed ticks can be stored in 70% ethanol pending identification and counting. The animal host is released at the site of capture after recovery from anesthesia. Gloves should be worn throughout all animal and tick handling operations. A veterinarian or qualified technician should be consulted on the proper usage of anesthetics administered to trapped animals.

Sampling sites for monitoring ticks should be selected in areas favoring ticks or are likely to receive heavy human visitation. A conscientious monitoring program is the basis of effective integrated pest management. Regular surveys should be done at all sites where ticks have been reported by park staff or visitors and at other locations that appear to be favorable tick habitats. Complete and accurate records of sampling sites and methods must be kept, so that the progress of tick populations and the effect of control measures can be gauged. After collecting the ticks, store them in rubbing alcohol or freeze in a plastic container to preserve them.

THRESHOLDS FOR TICKS

Mount (1981) proposed an arbitrary tolerance threshold of one tick/dry-ice sample, based on several years of study in recreational areas in Oklahoma. Mount and Dunn (1983) recommended that a count of 0.65 ticks per one hour of CO_2 exposure (dry-ice traps) be considered the economic threshold in lone star tick management (equivalent to one tick per visitor per day, based on the assumption that most human visitors to recreational areas will not spend more than one hour per day in tick habitats). This value may not be applicable to your particular situation and a suitable threshold level can be established by conducting regular CO_2 surveys and plotting tick counts against the numbers of tick bite complaints received. This will permit the selection of a complaint threshold level for each site surveyed. Treatment should be conducted to keep tick populations below the selected threshold; a lower "action" level should be selected to trigger treatment programs. The same technique is applicable to other species of ticks as well.

NON-CHEMICAL CONTROL OF TICKS

Education

Ticks are important disease vectors in many regions of the country. Park visitors and employees need to be aware of tick species and diseases present in their area, as well as personal protection measures that should be taken by anyone who will be in tick-infested areas. Parks should use interpretive displays to inform their visitors about ways to avoid contacts with ticks.

Biological Control

Several species of ants are known to feed on ticks. Recently, releases of the parasitic wasp *Hunterellus hookeri* have been made on several small islands on the New England coast. This wasp attacks *Ixodes dammini* and has been recovered from some of the release sites (Van Driesche, personal communication).

Habitat Management

Wherever possible, visitor activities should be directed towards areas that provide unfavorable habitat for ticks. Regular inspection of the park should be performed to determine when tick management needs to be initiated. The basic principles of management include isolation of susceptible domestic animals from known tick populations and rotation of pasture or run areas to reduce tick populations.

Removal of shrubs, trees, or tall grass can be useful in situations where it is consistent with National Park Service policy regarding use of the area. Dense shrub and tree cover and tall grass provide harborage for both ticks and their animal hosts. Removal of excess brush and shrubbery and clearing the canopy trees so that 50% to 80% of a management area is exposed to direct sunlight at any time are recommended control practices for walkways, parks, and landscaped grounds (Hair and Howell 1968). Grass should be kept below 6" in height to allow the penetration of sunlight and soil ventilation. Such techniques result in higher soil temperatures, lower humidities, and lower soil moisture, all of which lead to higher tick mortality. In one study, such techniques resulted in 75% to 90% control of different tick life stages of the Lone Star tick (Mount 1981). Mowing vegetation with a bush-hog rotary mower reduced adult deer tick populations by 70% in another study (Wilson 1986).

Controlled burning of habitat may reduce tick numbers and may be feasible in a park if it is consistent with a fire management plan. For example, burning tick- infested areas on Great Island, Massachusetts, reduced deer tick populations by 38% six months after the burn (Wilson 1986). However, the long-term implications of burning are unclear. Burning typically improves deer browse in the area; thus increased deer abundance may result in the movement of ticks back into the area.

Research has shown that high deer populations can lead to increased Lone Star and deer tick populations since there will be more hosts from which a blood meal can be obtained. Reducing the deer population may be a feasible tick management strategy in locations where it is compatible with National Park Service policy. This reduction has been experimentally demonstrated in Massachusetts (Wilson et al. 1988), although the decline in tick numbers may not correspond directly to the reduction in deer population. Managing deer populations by hunting, fencing, or environmental modification should be considered seriously before tick infestations become severe and should be done within state and local guidelines. Efforts at deer management should be done in coordination with state natural resources and wildlife department personnel.

Under unusually high tick population pressure it may be necessary to treat indoor areas. The major methods of nonchemical indoor tick management include regular inspection, elimination of animal (especially rodent) harborage areas, use of food and waste-handling procedures that minimize animal entry and harborage, and animal-proofing buildings. This includes sealing all holes in foundations and walls, and screening (with heavy gauge metal screen) aboveground windows, vents, and other openings through which animals may enter. A 18" perimeter border of gravel may prevent movement of ticks from grass areas into buildings. Cracks and crevices around the base of buildings should be sealed with caulk.

Recommended practices include frequent examination of clothing (preferably by another individual) and the body (after showering), destruction of collected ticks, and wearing protective clothing (e.g. coveralls with trouser cuffs taped to shoes, high- top shoes, socks pulled over trouser cuffs, long-sleeved shirts or jackets, or mesh jackets). Clothing should be light-colored so ticks may be easily seen.

Periodic surveys of potential or known habitats can reveal the presence of low- level tick infestations, thus indicating the need for application of management practices to prevent or retard further population increase.

CHEMICAL CONTROL OF TICKS

Outdoors

Insecticides or acaricides. Several insecticides and acaricides that provide effective control of tick populations in small infested areas. At least two treatments are required for control; one in the spring for adult and nymphal stages and the other in late summer for larval stages. Surveillance is necessary to determine times of application (see Monitoring section for techniques). Low to moderate infestations can usually be controlled by one spring and one late summer treatment; heavy infestations may need two or more treatments in the spring and again in late summer and early fall. Consult your regional Integrated Pest Management coordinator to determine pesticide choice and application rates.

Aerial dispersal of acaricides requires coordination with local, state, and sometimes federal officials. Chrlorpyrifos in a 14% granular formulation applied at 7 lb/acre has been used successfully in tick control by this method (Goddard 1989). The National Park Service, however, does not currently use this method due to extensive bird kills associated with chlorpyrifos.

Vegetation management by herbicides is another tick control option. It produces the same benefits as mechanical management of vegetation; i.e., reduced harborages for animal hosts of ticks, reduced soil humidity, and increased soil temperature, all of which are detrimental to tick survival. Management of vegetation by herbicidal and mechanical methods may not always produce comparable results; Hoch et al. (1971) found that herbicidal treatment of woodlots was not as effective as mechanical vegetation clearing in reducing the population of Lone Star ticks.

Personal Protection

Ticks can be prevented from attaching to the skin or clothing by the use of repellents. Schreck et al. (1980), reported that DEET, M-1960, and permethrin provided 81%, 95%, and 89% protection, respectively, against the Lone Star tick. Mount and Snoddy (1983) showed that the application of pressurized sprays of 20% DEET to the exterior of surfaces of clothing provided 85% protection against nymphal and adult Lone Star ticks and 94% protection against adult American dog ticks. Permethrin (0.5%) gave 100% protection against both species.

However, DEET and M-1960 have a disagreeable odor and can cause skin irritations. The most effective repellent/toxicant against all tick species available at present is Permanone(0.5% permethrin), which must be used as a clothing treatment; **Permanone is not intended to be sprayed directly onto the skin** (Goddard 1989). Permanone remains effective for at least 1 month on unwashed clothing. All pesticide-treated clothing **must** be washed separately.

Indoors

Sites such as crevices, baseboards, trimming, furniture, ceilings, floors/carpets, walls behind pictures, bookshelves, and drapes should be spot-treated as needed. Crack and crevice treatments should be done with residual dusts or silica gel. This is the most effective way to use pesticides in a building. Fumigation does not work well in buildings because ticks can readily re-enter through doorways or windows.

SUMMARY

For outdoor areas, habitat reduction by mechanical removal of excess brush and overstory and regular mowing of grass 6" or less in height is recommended. Regular CO_2 or drag surveys of likely tick habitats will indicate locations where treatment is required. If nonchemical measures prove ineffective, registered herbicides (for vegetation management) or acaricides (for direct kill) may be needed.

Animal-proofing park buildings through the use of exclusion techniques should eliminate indoor tick habitats and reduce the chance of future infestations.

Recommended procedures for protection of park personnel and visitors include frequent examination of the clothing and body of any person travelling through tick habitats, wearing protective clothing, and the use of clothing and/or skin-applied tick repellents.

Information should be made available to park visitors concerning known tick habitats within the park, personal protection techniques, and tick removal techniques.

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Turfgrass Insects

This module is intended to serve as a source of basic information needed to implement an Integrated Pest Management program for turfgrass insect pests. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

While many species of insects damage turfgrass in the United States, this package describes in detail the life histories and management of those which have been found to be frequent problems within the National Park System. Photographs and further information concerning these and other turf pests can be found in publications such as Niemczyk (1981), Daley (1975), Shetlar (1982), Converse (1982), or Tashiro (1987).

MONITORING FOR TURF INSECTS

This section discusses monitoring techniques and strategies applicable to all turf insects. Information on monitoring and thresholds for specific turf pests is given in the section of this module that deals with that pest. Most detection and sampling methods can be classified as either active or passive techniques. Both have the capability to help in **predicting** pest problems or **quantifying** existing damage and pest infestations. The most popular and efficient active sampling method involves visual inspections (scouting). Passive systems use either light, pheromone, or mechanical traps that require fewer site visits per season. The trade-off between visual and passive systems is accuracy. Traps will tell you if an adult insect is present in the sampling area, but will not show if it is causing injury or how extensive the population is. These can usually only be determined by some type of visual inspection. The best monitoring program combines both visual inspections and a variety of passive methods.

Regardless of the sampling methods used, the trade-off becomes time for accuracy v. cost. Scouting cost and time can be reduced and accuracy maintained if monitoring is concentrated on key pests in key locations. Key pests are those responsible for major turf losses at a particular site. Key locations reflect the behavior of the key pest to habitually select and damage the same turf areas over time. For example, Japanese beetles are most likely to be found in turf areas which receive full sun, so these areas are considered key locations for Japanese beetles. Key areas are sites that are unique because of aesthetics, rarity of plant material, or historic value. These areas may require lower damage thresholds to meet the expectations of visitors.

Monitoring Based on Key Locations

Predetermined key locations should be intensely monitored to detect the first occurrence or first damage. Many turf insect pests require warm, moderately-dry turf conditions for optimal development. For example, chinch bugs prefer the full sunlight of southern and eastern exposures. Japanese beetle adults lay more eggs in well- watered, sunlit turf areas as compared to dry areas, and billbugs will lay more eggs close to driveways and sidewalks than in the open turf. In southern regions areas first damaged by migrant adult mole crickets in the spring will become the most severely damaged in late summer. These examples demonstrate the need for understanding the biology and behavior of turf pests in relation to the site.

Other key locations that require special monitoring are areas regularly infested every year. Accurate records, or an experienced manager's memory, are important in identifying these areas. If historical documentation is unavailable, pest damage, site, weather, and turf characteristics must be correlated. Over time these sites will become evident. This type of detailed factual record-keeping will help you select the appropriate management tactics. Once a key site is identified, modification of the habitat or vegetation can reduce the reinfestation problem and, in the long-term, reduce pesticide usage. In many situations the elimination of turfgrass or replacement with a better adapted turfgrass species will solve the problem.

Sampling Techniques

Visual Inspection

These methods are the quickest, most accurate, and most frequently used technique for detection of turf insect problems. The observer, however, must have a wide background or training in turf management, disease management, and insect pest management in order to make the correct diagnoses. Detection of insect damage or the prediction of pest outbreaks also depends on repeated observation of insect adult activity as well as recognizing small changes in plant appearance that are diagnostic of pest injury. The frequency of scouting visits depends on the pest complex, visitor expectations, thresholds, and costs. Typically a weekly or bimonthly schedule in spring and summer is sufficient, while a monthly schedule is acceptable after mid-August into the fall. The following examples show situations and conditions where direct observations are essential to a successful integrated pest management program.

Spot sampling. Trained individuals can quickly make accurate observations and pest counts. A 30-second spot count per square foot sample in 20 or so turf locations should provide information on the scope of damage, stages of pest present, and population estimates. Spot counts require searching the thatch and root zone thoroughly. All pest species can be detected with this method. Although accurate, visual inspections only reflect a population response to the environmental and site conditions at one point in time. For example, the weather may be cool or excessively wet at the time of sampling. These conditions tend to slow down insect development, daily movements, and response to flushing agents. Generally samples taken under extremes in temperature and moisture tend to underestimate populations. This can be avoided by increasing the number of sample dates each month or avoiding sampling during weather extremes. Supplemental sampling with traps should provide a better population estimate because the counts reflect an average over longer periods of time. Traps collect samples every day regardless of environmental conditions, personal discomfort, and human variability. Scouting is usually done during the daylight hours but during hot weather insects may not be active until late evening so traps may alert you to unseen problems. If a problem is identified via a spot count, one of the following methods will provide a better estimate of threshold populations than spot sampling.

Irritant sampling. This method is more accurate than the 30 second spot counts, particularly where insects are hidden in thick thatch or cracks in the soil. The irritants are only recommended for sampling highly mobile insect pests (Niemczyk 1981). Irritants will not expose soil pests such as white grubs and billbug larvae. It is most effective when turf is mowed or clipped before making observations.

Species living in the thatch such as sod webworms, cutworms, chinch bugs, and billbug adults respond to irritant agents. Flushing mole crickets from the soil is also possible, but accuracy may diminished due to of variations in thatch thickness, soil temperature, soil moisture, and depth of feeding activity. To flush with soap in non- thatch situations, use 1 ounce of liquid detergent in 1 gallon of water per yd^2.

Use of the irritant method requires both a through soaking of the thatch layer or soil and close observation in order to detect the excited insects. Most insects will exit the thatch within five to six minutes and move out of the sample area quickly. One person may have difficulty observing this activity over an area of 1 yd^2; the area can be reduced to 2 ft^2 if necessary. To take an irritant sample, a circular metal retaining frame which is 27" across by 6" high is forced into the soil through the thatch layer and filled with 4 ounces of liquid detergent in 4 gallons of water. Count the number of insects which float to the top after ten minutes.

Flotation sampling. This is used primarily for estimating chinch bug populations. A 1 or 2 pound open-ended coffee can or a specially-made cylinder with handles is forced into the soil through the thatch layer and 3"-4" inches of water poured inside. If the water recedes more water must be added to maintain that level. All stages of the chinch bug, as well as the principal predators such as the big-eyed bug, should float to the surface within 5-10 minutes.

Although very accurate, flotation sampling is also time-consuming. This method confirms chinch bug infestations or determines the extent and population density of infestations.

Soil sampling. Although the most difficult and time-consuming of all the visual sampling methods used in turf monitoring, soil sampling provides the most accurate method for determining white grub and billbug larval population densities. Samples are initially taken in areas where turf insects have been or are expected to be a problem, including sunny areas with adequate moisture, areas where insect damage is visible, and areas where previous treatment was needed. The location and severity of grub infestations are detected by a circular sampling pattern. Once areas of grub infestation are located, samples are taken in a circular pattern which expands out from the initial site. Continue to sample outward from the initial site until grub counts become low enough to no longer be a concern.

The 1 square foot sod spade method is the least destructive but most time- consuming way to sample turf. A 1 ft^2 sample is cut on three sides to a depth of 3" to 4" and the sod square is folded back to expose the soil. The soil is then broken apart, the grubs counted, and the soil returned to the hole. The grub counts are recorded and the sod flap is returned on top of the loose soil. This method is more accurate than the sod cutter but many samples are required for a good population estimate. Adjustments in the depth of cut can be made in the fall and spring when grubs are more widely distributed throughout the soil profile.

A standard golf course cup cutter allows more samples to be taken, increasing the sampling accuracy relative to the spade technique. The standard cup cutter is 4.25" in diameter; this one-cup cutter grub count can be converted to a square foot basis by multiplying by 10.15. Begin by taking soil cores to a depth of 3"-4" at key locations; do more extensive sampling if grub activity is detected.

A sod cutter produces the quickest soil sample. During the summer and early fall, population estimates are made by removing a series of 1"-2" thick sod slabs and counting the unearthed grubs. Unfortunately some grubs will be missed if they feed at the thatch level or well below the 2"-3" level, particularly species that move rapidly up and down in the soil profile in response to moisture, such as the European chafer, green June beetle, and oriental beetle. This method is less accurate in the late fall and early spring because grubs are moving up and down in the soil profile.

Passive Sampling Techniques

Light traps, mechanical traps, and pheromone traps are best used in a park-wide integrated pest management program. Although less accurate than the visual method, these traps are still useful in monitoring a pest's presence, especially yearly and seasonal population fluctuations. They also aid in scheduling peak scouting activities.

Blacklight traps. These systems can collect large numbers of sod webworm and white grub adults. To date there is no way of estimating the damage potential or the resulting larval population from these adult counts, but information on adult activity obtained from these traps is useful in determining when larvae will be active. The high cost of $200-$300 per trap often makes this option prohibitively expensive.

These traps will also provide notice of the first occurrence of a pest and delineate the species distribution over a large geographic area. Determining the relative abundance of species and risk assessment of damage from one year to another at the same sampling site is another important use of light traps.

Pheromone traps. Pheromones are chemicals which are emitted by an organism to communicate with other members of its species. The most widely used pheromone trap in turf integrated pest management programs is the Japanese beetle trap. This trap uses a floral lure and female sex pheromone. The high price per trap and the excessive number required per acre to significantly reduce the adult beetle populations limits the use of trapping of beetles as a management tactic. Trapping can be used as a monitoring tool to detect the buildup of populations and to monitor variations in populations between geographic locations or from one year to the next. Most Japanese beetle traps utilize both the floral and sex pheromone lures. The sex lure increases the number of male beetles collected by 10-40%. The traps should be used without the sex lure component when they are used for population monitoring so trap catches will more accurately reflect the normal 50:50 sex ratio. Monitoring the females is more important because they lay the eggs that give rise to the damaging grub population. Additionally, some parasites that attack the adult beetle, such as the Winsome fly (*Istocheta aldrichi*), can be easily collected from trapped adult beetles and distributed into new communities or geographic locations.

Daily beetle trap catches will be strongly influenced by temperature, rain, distance from host plants, soil type, groundwater levels, and natural enemies. Using several traps at each sampling location will help reduce the effect of this variability on population estimates.

Pheromones for several other species of annual white grubs are now being tested and should be on the market within a few years. Otherwise, lures for species such as black cutworm, true armyworm, and fall armyworm are now available for use. Unfortunately, little progress is being made on the sod webworm species other than the cranberry girdler, *Chrysoteuchia topiaria* (Zell). This sampling tool will become more important in the near future when additional pheromone systems are marketed for turf insects.

Pitfall traps. These are primarily used to monitor billbugs, mole crickets, chinch bugs, and other highly mobile arthropods. The basic design is a small hole or pit lined with a slippery-sided container with the bottom filled with a killing liquid such as soapy water or alcohol. Small pin holes should be punched into the bottom of all the cups to allow water drainage. Although the trap-line setup takes time, daily, season-long inspection is quick. This trap is best utilized for monitoring of first occurrence for the length of the adult activity period.

Data collected from insect traps becomes more meaningful if used in conjunction with degree-day heat unit life history models. These models measure the amount of heat over a certain minimum temperature (usually 50F) which a site has accumulated after a given date (usually March 1st). For example, if the minimum temperature during a 24-hour period is 60F and the maximum is 70F, the average is 65F. This is 5F higher than the base temperature of 50 F, so the degree-day accumulation for that period is 5. If the same average temperature were to occur each day that week, the degree-day accumulation at the end of the week would be 35. For certain insects, we know how much heat needs to accumulate before they will emerge from their overwintering stage. By keeping track of degree-day information we can accurately predict when these insects will emerge. Such a heat-unit model is now available for the bluegrass billbug.

Additional Benefits of Monitoring

Prediction of outbreaks. Regardless of the monitoring method, observations of adult activity of turf insect pests can help in predicting problems. High populations infer high risk of damage later in the season. Generally, adult females will be active and laying eggs two to four weeks before the immature stages start to cause noticeable damage. For example, if one billbug adult is observed per minute of observation, sufficient egg-laying and subsequent larval damage will occur that may require preventative treatments (Niemczyk, 1981). Observations in Ohio (D. Shetlar pers. comm.) showed that if billbug pitfall traps caught two to five adults per day during the peak egg-laying period, moderate turf injury could result in July. More severe losses could occur if counts exceed seven to ten per day over several days.

Outbreak detections in adjacent sites. Detection of injury or pest activity in areas adjacent to the key location may indicate that these areas run increased risk of developing a problem in the future. These key locations should be monitored more frequently than other key locations. This often occurs with chinch bugs and billbugs, since they rarely fly but tend instead to walk slowly from one area to another.

ANNUAL WHITE GRUB SPECIES COMPLEX

White grubs are the larvae of many species of beetles, most of which belong to the family Scarabaeidae. Although the adults differ from one another in appearance and life cycles, the grub stage of all species are similar in appearance. Fully grown larvae are 1/2" to 3/4" long, white to grayish, with brown heads and six legs. Refer to Tashiro (1987) to see the relative sizes of grubs. They generally assume a C-shaped position while in the soil. Grubs can be identified to species on the basis of the raster setal hair pattern found on the underside of the last abdominal segment. These are illustrated in Tashiro (1987). The raster is easily seen using a 10x magnifier.

Although numerous species of grubs cause turf damage, five key species are described in detail in this module; the Japanese beetle, northern and southern masked chafers, European chafer, and oriental beetle. The life history patterns for these species are very similar, although the peak emergence period for adults varies somewhat and under drought conditions some species will delay adult emergence until rains or irrigation moisten the soil profile. The Japanese beetle life cycle is typical of all the key species with annual cycles.

JAPANESE BEETLE
IDENTIFICATION AND BIOLOGY

The adult male beetle is 3/8" long; the female is slightly larger. Roughly oval in outline, the head and prothorax of Japanese beetles are greenish bronze. The wing covers are brownish bronze with green along the sides and center. Japanese beetles have twelve white tufts of hair are present along the sides of the abdomen and at the tips of the wing cover, and plate-like antennae. The legs are long, with heavy claws. The round eggs become oval after absorption of soil moisture, measuring less than 1/16" across. Larvae molt twice; a third instar grub can be up to 1" long. The head is brown, with large, brown-black mandibles.

The Japanese beetle is an Asian native that was first reported in the United States at Riverton, New Jersey, in 1916. It is common in all states east of the Mississippi River except Florida, Mississippi, and Wisconsin. It has also been found in Missouri, Minnesota, and California. Because of the ease of shipping grubs with nursery stock and soil, this species could potentially be found anywhere in the United States, including Hawaii and Puerto Rico. Adults are highly mobile and frequently hitch rides in airplanes and cars. National Park Service lands are at particularly high risk for infestation because of the unrestricted public access.

Adult Japanese beetles feed on more than 300 species of plants including many trees, ornamental shrubs and vines, fruits, flowers, vegetables, and weeds. Adults feed on foliage and fruit, while larvae feed on roots, especially those of grasses, vegetables, and nursery plants. Females prefer to lay eggs in warm, moist soil where turfgrass is exposed to full sunlight.

MONITORING AND THRESHOLDS FOR JAPANESE BEETLES

Larvae feed on grass roots and may move up into the thatch layer after consuming the total root system. In Kentucky bluegrass, populations of 6-8 grubs per ft^2 may kill turf during August-October when it is drought stressed, while under the good growing conditions in spring counts of 10-15 per ft^2 may cause no damage symptoms. Clump grasses such as tall fescue tolerate higher summer populations but in mixed tall fescue/bluegrass sod, grubs will selectively kill out the bluegrass component.

This root feeding injury gives the turf a spongy feeling when walked upon; after heavy feeding the roots are severed and the sod easily rolled back to observe the grubs. Without irrigation the turf will turn brown, die, and usually not recover. Some improved varieties of Kentucky bluegrass with extensive underground stem systems have better recovery prospects.

Generally, treatments are recommended when grub populations exceed 6-8 per ft^2. However, moles and skunks may also destroy turf; because of these small mammals, treatment thresholds can range as low as 4-5 grubs per ft^2. In this situation the grubs do not kill the turf; the animals destroy the sod when they dig for the grubs. Cultural conditions such as turf type, amount of irrigation, or time of year may also affect treatment thresholds. If bluegrass turf areas are frequently irrigated during the July-August stress periods, populations as high as 20-25 grubs per ft^2 may not produce damage until the irrigation stops. Kentucky bluegrass, perennial rye, and fine fescue grasses are the most sensitive to grub feeding damage. Tall fescue and warm-season species such as zoysia and bermuda can tolerate moderate to heavy populations during the hot dry conditions that kill Kentucky bluegrass lawns. Mixed populations with the masked chafers and other grub species are common throughout the species range.

NON-CHEMICAL CONTROL OF JAPANESE BEETLES

Biological control

Biological control options include milky disease (*Bacillus popilliae*), new *B.t.* strains, parasitic wasps and flies, and parasitic nematodes.

Milky disease provides the first option in the mid-Atlantic states for low-to- moderate maintenance turf situations. This bacterial agent is long- living, species- specific, self-perpetuating, easily-applied, and best adopted to the mid-Atlantic region. Success in New York and New England areas has been limited due to the average cooler soil temperatures and shorter grub season.

This bacterial disease suppresses only the Japanese beetle grub and requires the presence of moderate grub populations in order to increase the soil spore counts and spread the disease to untreated areas. Spores can remain viable for 20-30 years once established in a lawn. However, at the economical rates of application recommended by the manufacturer, effective control may require a two-to-four year establishment period. The granular formulations have not been proven effective to date. The spore dust product is recommended for newly-established turf areas. Areas with ten or more years of turf coverage may already have naturally infected soils. Remember that this disease works most efficiently when grub populations are moderate to high. This situation usually occurs in new housing developments. Some occasional damage can be expected regardless of the spore levels in the soil and age of the turf.

Milky disease is not compatible with insecticides because they kill the grub population that is required to increase and maintain the soil spore counts. Fortunately the spores will remain viable after an insecticide treatment and continue to increase in later years when the grub populations return.

Natural Microbial Control

Japanese beetle grubs are infected by many species of bacteria, fungi, protozoans, and nematodes. Few of these have commercial possibilities but together they constitute one of the major grub population regulators in established turf pastures and meadows. In many older communities problems rarely occur, particularly where turf has been established for eight to ten years or more. The highest risk of grub damage occurs in new lawns two to five years of age or newly-disturbed sites reseeded to turfgrass. New turf areas are often developed from farm fields or wood lots that lack a grub pathogen complex. Pathogens have not developed at these sites because the Japanese beetle and other important grub species do not occur naturally in these habitats. Once grub populations become established after several years, key pathogens and other biological agents also become established and thus help maintain low grub populations.

In the past, *B.t.* products did not suppress grubs but recent research reveals that both newly discovered and bioengineered strains can provide this missing capability. These new strains show efficacy against the Japanese beetle and masked chafers. Presently the research appears very promising, but commercial labeling is not expected before 1995. At that time, new labeling for *B.t.* mixtures that control grubs, armyworms, cutworms, and sod webworms should be competitive with insecticides and parasitic nematodes.

Wasp and Fly Parasites

The most effective wasp parasites are the Tiphia wasps. Introduced from Japan, *Tiphia popilliavora*, and Korea, *T. vernalis*, these species attack the grub stage in thatch or soil. Historically established throughout the mid-Atlantic region, populations of both species are now low with only scattered pockets of high rates of parasitism. These two species are most effective in areas that have had high populations of grubs (10-30 square foot) over long periods of time. They do not appear to be effective at low population densities which are typical of 90% of our turf areas today. However, they should be introduced into newly-infested areas around the country.

When the adult wasps are abundant, people mistakenly view these low-flying insects as another type of honeybee or yellowjacket, ready to attack and sting. In reality, they are not aggressive and rarely sting even if handled. People throughout Japan accept these wasps without any fear of being attacked.

The only major adult Japanese beetle parasite is a fly imported from Japan, *Istocheta aldrichi*, sometimes referred to as the Winsome fly. This species prefers adult female beetles and deposits an egg just behind the head on the prothorax. The egg hatches within 24 hours and the maggot enters the beetle body. The adult beetle eventually is consumed and dies within six days. After the adult falls to the ground or buries itself before death occurs, the parasitic fly larva pupates and remains in the soil until the following year.

This fly parasite is limited in distribution to eastern New York and the New England states. USDA and state entomologists made attempts over the past eight years (1982-1990) to reintroduce this New England fly strain into other states (Ohio, North Carolina, Maryland, Virginia, and Kentucky). This effort will continue for another few years. To date this fly has not been recovered in any of these new release sites. However, there may be areas that have localized, undiscovered natural populations. Because the fly is a poor disperser these areas should be explored and parasites collected and moved to new Japanese beetle infestations.

Parasitic Nematodes

The newest grub biological control agents are parasitic nematodes. These nematode species are selected for their ability to enter the soil, seek out all species of grubs, and quickly parasitize them. Research to date shows that they must be applied when the soil moisture is relatively high for optimal results. Several experimental applications of nematodes at rates of 1 to 5 billion nematodes/acre confirm that the percentage control has been generally good, with the upper ranges near the levels expected with the best soil insecticides. Under optimal conditions one application per season for grub control should be sufficient.

A commercial product with the nematode *Steinernema carpocapsae* is marketed under the trade names BioSafe, BioVector, and Exhibit. Recent research trials throughout the northeast United States indicate a wide range of efficacy with this species of nematode. The reason for such poor results appears to be a combination of factors, such as product quality, cool soil temperatures at time of application, and extremes in soil moisture levels. Presently *S. carpocapsae* is not recommended for grub control in turf because this strain appears to be poorly adapted for soil insect control. New product species with *S. glasseri* and *S. feltae* and *Heterorhabtis sp*. appear to be better adapted to search out grubs and other soil insects.

Fortunately the interaction problems with microhabitat and environmental factors can be resolved with new genetic selections from these new species. To date research continues with several companies testing these better-adapted strains. These selections should reach the market in the mid-to-late 1990s.

Cultural Control

Soil moisture levels are very important in assessing damage risks in turf. Japanese beetle eggs must acquire moisture from the soil in order to develop and hatch. Research has shown that soil moisture levels less than 10% are lethal to the eggs and newly-hatched grubs. Irrigated areas are also very attractive to egg-laying females, and they ensure good larval survival. If daily irrigation is maintained during these grub outbreaks, Kentucky bluegrass turf can sustain 20 or more grubs per square foot without showing injury. However, once the water is withdrawn, even briefly, the turf dries out and dies.

Turfgrass species and variety selection can greatly influence the susceptibility to grub damage. Perennial rye, bentgrass, Kentucky bluegrass, and fine fescue species are the most susceptible to severe damage, whereas the warm-season grasses like zoysia and bermuda simply outgrow damage and tolerate higher densities of grubs. Tolerance of tall fescue varieties is intermediate to that of bluegrass and warm-season grasses.

CHEMICAL CONTROL OF JAPANESE BEETLES

Consult your National Park Service regional Integrated Pest Management coordinator for pesticide recommendations for Japanese beetle control in your area.

NORTHERN AND SOUTHERN MASKED CHAFER IDENTIFICATION AND BIOLOGY

The southern masked chafer (*Cyclocephala lurida*) and the northern masked chafer (*Cyclocephala dorealis*) are small, yellow-brown beetles 1/2" long with darkened black to chocolate brown areas (masks) between the eyes. The mask becomes lighter in color toward the mouthparts and helps separate these two species from the numerous other similar-appearing May and June beetles. The grubs can be identified by their unique rastral pattern, which is illustrated in Figure 30 on page 118 of Tashiro (1987). Both species have a life cycle similar to the Japanese beetle, except that the adults fly at night and don't feed. Females prefer the same sites as Japanese beetles for egg-laying. Grubs generally feed in upper 1"-3" of the soil profile but move up into the thatch layer for moisture when turf is moisture-stressed.

A broad overlapping distribution occurs in the mid-Atlantic states. Because the grub feeding damage and life histories are similar, both species are frequently referred to as masked chafer grubs. A grub species shift is occurring in many mid-Atlantic areas where masked chafer grubs overlap with the Japanese beetle; reasons not fully understood, urban neighborhoods that previously hosted 80-100% Japanese beetle populations now host to a predominately masked chafer population. Selective natural control by milky disease and other Japanese beetle specific pathogens may be part of the explanation for the shift, as masked chafers are not affected by the Japanese beetle milky disease.

MONITORING AND THRESHOLDS FOR NORTHERN AND SOUTHERN MASKED CHAFER

Adult masked chafers do not feed and grub damage is identical to Japanese beetle. Populations of 10-15 grubs per square foot can severely damage turf. The adults of both species can be monitored with blacklight traps. The southern species flies just after sundown and activity stops around 12:00 pm. The northern species starts its flight activity at 2:00 am and continues until sunrise. Both species are highly attracted to irrigated lawns during the dry weather and both flight and egg-laying activities will increase significantly after rainstorms.

Masked chafer grubs also feed on organic matter. This results in higher population thresholds for these species. Potter (1982) indicated damage thresholds were 9-10 grubs per square foot for moisture-stressed turf and 15-20 per square foot for non-stressed irrigated turf. Similar to the Japanese beetle, masked chafers frequently intermix with other annual white grub species.

Unlike the day-flying and feeding Japanese beetle adults that can alert turf managers to possible future grub problems, the masked chafer adults fly at night and cause no feeding damage, so turf managers often have no warning of an outbreak. Masked chafer grub injury is often misdiagnosed as Japanese beetle feeding.

NON-CHEMICAL CONTROL OF NORTHERN AND SOUTHERN MASKED CHAFER

Biological Control

A new strain of milky disease specific for *Cyclocephala* species has been discovered and looks promising but commercial production may take several years. The Japanese beetle milky disease will not suppress this pest. The parasitic nematodes used to control Japanese beetles will also suppress masked chafers.

Cultural Control

The same moisture conditions and turfgrass varieties that reduce Japanese beetle damage will also affect the masked chafers. The wide use of milky disease to control the Japanese beetle has allowed this species to increase its populations because of the lack of competition from the Japanese beetle.

CHEMICAL CONTROL OF NORTHERN AND SOUTHERN MASKED CHAFER

Consult your regional National Park Service Integrated Pest Management coordinator for information on chemical control of these pests in your area.

MAY/JUNE BEETLES IDENTIFICATION AND BIOLOGY

May or June beetle (*Phyllophaga* spp.) adults are 3/4" - 1 3/8" long, stout, shiny red-brown to blackish-brown. The antennae have three plate-like segments forming a club-like structure at a right angle to the other segments. The head, prothorax, and wing covers usually have no distinguishing markings or grooves. Larvae are white with brown heads, have large jaws, and have an elongated raster featuring two parallel lines of hairs, oriented front to rear on the segment. The rastral pattern is pictured in Figure 30 on page 118 of Tashiro (1987).

These species prefer open woods, meadows, lawns, grasslands, cultivated fields and ornamental plant beds. Grubs feed on organic matter and plant roots. The adults of several species cause defoliation of ornamental and shade trees.

These beetles have a two-to-three year life cycle, depending on the species. Eggs are deposited 1"-8" deep in the soil in late spring. The eggs hatch in about three weeks and young larvae begin feeding on roots and decaying vegetation. In the fall, they migrate down into the soil, where they overwinter. The three-year cycle species resume root feeding in the following spring. After a summer of damaging feeding, they hibernate deep in the soil over their second winter, and then rise to near the surface to feed again until about June. Pupation then occurs in a hollowed cavity in the soil. The new adults emerging from these pupae (in about three weeks) remain in the hollow cavities through the following winter and only emerge the following May or June, when feeding, mating, and egg-laying occur.

Although these insects require two to three years to complete their development from egg to adult, adults are present every year because three different broods produce adults in different years. Brood A (with adults to be seen in 1986, 1989, 1992, and so on) produces the greatest damage. Next in importance is Brood C (adults in 1985, 1988, 1991, etc.). Brood B (adults in 1987, 1990, 1993, etc.) is of least importance, since it consists of the fewest individuals (Davidson and Lyon 1979).

Adults fly and feed during the night but hide in soil or sod by day. Trees with new tender spring growth are most susceptible to adult defoliation. Although named May/June beetles, some species emerge in April.

Larval damage to turf is similar to that caused by Japanese beetle grubs. No thresholds are available in the literature. Limited studies in Maryland show that five to seven grubs per square foot may damage turf under drought stress conditions.

NON-CHEMICAL CONTROL OF MAY/JUNE BEETLES

Biological Control

There have been several reports of *Bacillus popilliae* infecting May/June beetles but it is considered a rare occurrence and a minor influence. Since these species are native to American soils, many species of nematodes, bacteria, and fungal pathogens have been recovered throughout their range.

Similar to the diverse microbial pathogen fauna, numerous species of parasitic wasps and flies attack the adult, grub, or pupal stages. These natural control agents appear to maintain populations below aesthetic damage levels in most areas.

CHEMICAL CONTROL OF MAY/JUNE BEETLES

Insecticides labeled for Japanese beetle also control these species. Consult your National Park Service regional Integrated Pest Management coordinator for information on pesticide selection and timing in your area.

BLACK TURFGRASS ATAENIUS AND *APHODIUS GRASSARIUS*

IDENTIFICATION AND BIOLOGY

Black turfgrass ataenius (*Ataenius spretulus* Haldeman) and the related dung beetle (*Aphodius granarius* (L)) adults are very similar in appearance. Adult beetles are red- brown, darkening to black with aging. Both are roughly oval in shape, and may be up to 3/8" long. Eggs are round, very small, and laid in clusters of 8-12 in the thatch. Grubs are no longer than the adults and resemble other grubs in color, but can be separated by rastral patterns. See Figure 30 page 118 in Tashiro (1987) for an illustration of the rastral patterns. These species have been reported from every continental state except Nevada and Montana.

Both species prefer the same turfgrass microhabitats and frequently form mixed populations. As members of the dung beetle sub-family, the females are highly attracted to dung, compost, piles of decaying grass clippings, and turf thatch. Pest level populations are primarily restricted to golf courses having a combination of high percentage of annual bluegrass and high levels of organic matter in the soil profile. Under these conditions grubs can damage fairways, tees, and greens.

Although these two species are ubiquitous in lawns, meadows, and woodlands, they rarely attain pest status in these habitats. In all habitats, root feeding may be secondary to organic material consumption. Damage to annual bluegrass, Kentucky bluegrass, bentgrass, and fine fescues has been reported, but 80% of the severe damage occurs in the annual bluegrass turf.

There are one to two generations of black turfgrass ataenius per year, depending on climate. There are two generations each year in Ohio, but only one in states farther north (Niemczyk 1981). Adults overwinter 1"-2" below surface in well-drained soils, or under leaves, pine needles and other debris near wooded areas. They emerge in April, mate, and females burrow into the turf to lay their eggs. During May and June, clusters of 8-12 eggs are laid in the thatch or the top 1/2" of soil. Larvae are present from late May to mid-July in thatch and soil. From late June to mid-July, mature larvae burrow 1"-3" inches into the soil, pupate, and emerge as adults in July and early August. These first generation adults lay eggs in July. The second generation larvae complete development and pupate in late August or September. The new adults emerge and seek final overwintering sites during the fall. The life cycle for the *Aphodius* species is not well-documented. One or two generations are reported with the first occurring one to two weeks earlier than the black turfgrass ataenius.

MONITORING AND THRESHOLDS FOR BLACK TURFGRASS ATAENIUS

Damage from black turfgrass ataenius is similar to Japanese beetle injury. The turf shows localized dry or wilted spots that coalesce over time into large, brown, dead areas. The roots are severed and the dead turf is easily rolled back to expose the feeding grubs. Turf will not recover from severe damage and losses will continue under irrigation.

Peak populations in Maryland golf course fairways range from 150-300 per ft^2. An infestation of 30-40 grubs per ft^2 will kill annual bluegrass and bentgrass. Kentucky bluegrass and tall fescues are tolerant of similar populations.

NON-CHEMICAL CONTROL OF BLACK TURFGRASS ATAENIUS

Cultural Control

Replacement with other more tolerant grass species such as perennial rye, Kentucky bluegrass, zoysia, bermuda, or tall fescue is highly recommended. Bentgrass greens still remain susceptible but the attractiveness to the female is greatly diminished with the elimination of annual bluegrass from the golf course. Elimination of grass clippings piles is also advisable.

Biological Control

A naturally occurring milky disease similar to the Japanese beetle milky disease has become established in most areas east of the Mississippi River. Mortality in the range of 30%-90% has been maintained for seven to ten years on many courses that were devastated in the late 1970's and early 1980's. This milky disease is not produced commercially but spreads by natural agents, mostly birds.

CHEMICAL CONTROL OF BLACK TURFGRASS ATAENIUS

The preference of these insects for high levels of soil organic matter poses a problem for chemical control of these insects because pesticides tend to bind to the thatch and fail to thoroughly penetrate the soil profile. The accumulation of a thick thatch layer also repels irrigation water during the summer drought periods. These combined conditions of thick thatch and high levels of organic matter make grub control extremely difficult with the best insecticide regardless of irrigation levels. Under these extreme circumstances many resource managers prefer to control the adults in the spring before they lay eggs in the thatch. The females entering the thatch to lay eggs are quickly controlled by the insecticide.

To determine when adults will begin laying eggs, visually inspect golf greens for adult beetles.

The egg-laying period for the first annual generation corresponds to the full-bloom periods of Vanhoutte spirea and horse chestnut, and the first-bloom periods of the black locust. Second generation egg-laying coincides with the blooming of the rose- of-Sharon (*Hibiscus syriacus*). Observation of these indicators may be used as a basis for applying controls, but monitoring adults with a blacklight trap is a more accurate method.

Use of herbicides to eliminate *Poa annua* is also an essential component of a control program for these beetles.

GREEN JUNE BEETLE

IDENTIFICATION AND BIOLOGY

The green June beetle (*Cotinus nitida*) adult is usually 3/4"-1" long, and 1/2" wide. The top side is forest green, with or without lengthwise tan stripes on the wings. The underside is a metallic bright green or gold, bearing legs with stout spines to aid in digging. In the mid-Atlantic region the names "June bug" and "June beetle" are commonly used for this insect, while they are called "fig eater" in the southern part of their range. They should not be confused the familiar brown May or June beetles that are seen flying to lights on summer nights. The green June beetle adult flies only during the day.

The larvae are white grubs often called "richworms" because they prefer high levels of organic matter for food. With three growth stages, the beetles develop similarly to the other annual scarab species. Their body lengths reach 1/4", 3/4", and 2" respectively. The larvae have stiff abdominal bristles, short stubby legs, and wide bodies. One unique characteristic of this grub is that it crawls on its back by undulating and utilizing its dorsal bristles to gain traction. Other typical white grubs, like the Japanese beetle grub, are narrower, have longer legs, crawl right side up, and when at rest assume a c-shaped posture. This species is native to the eastern half of the United States and overlaps with *Cotinis texana* Casey in Texas and the southwestern United States.

The adults generally do not feed but occasionally become pests of fruit. Any thin- skinned fruit such as fig, peach, plum, blackberry, grape, and apricot can be eaten. The principal attraction is probably the moisture and the fermenting sugars of ripening fruit. They occasionally feed on plant sap. In turf situations egg-laying females are attracted to moist sandy soils with high levels of organic matter. Turf areas treated repeatedly with organic fertilizers, composts, or composted sewage sludge become more attractive to the female.

The grub feeds on dead, decaying organic matter as well as plant roots. This species is commonly associated with both agricultural crop and livestock production areas as well as urban landscapes. Field-stored hay bales, manure piles, grass clipping piles, bark mulches, and other sources of plant material that come in contact with moist soil provide prime microhabitats preferred by both the female for egg-laying and the migrating third instar grubs.

The green June beetle completes one generation each year. Adults begin flying in June and may continue sporadically into September. On warm sunny days, adults may swarm over open grassy areas. Their flight behavior and sounds reassembles that of a bumble bee. At night they rest in trees or beneath the thatch.

After emerging, the adult females fly to the lower limbs of trees and shrubs and release a pheromone that attracts large numbers of males. Frequently, males repeatedly fly low and erratically over the turf trying to locate emerging females. After mating, females burrow 2"-8" into the soil to lay about 20 eggs at a time. The spherical eggs are white and almost 1/16" in diameter.

Most eggs hatch in late July and August. The first two grub stages feed at the soil thatch interface. By the end of September, most are third instar larvae and these large grubs tunnel into the thatch layer and construct a deep vertical burrow. The grubs may remain active into November in the mid-Atlantic region. In the more southern states grubs may become active on warm nights throughout the winter. In colder areas they overwinter in burrows 8"-30" deep. The grubs resume feeding once the ground warms in the spring and then pupate in late May or early June. The adults begin emerging about three weeks later.

MONITORING AND THRESHOLDS OF GREEN JUNE BEETLES

The green June beetle grub also differs from other white grubs in their feeding behavior. Damage to turf occurs as a result of their unusual habit of tunneling as well as root feeding. Smaller stage grubs tunnel horizontally in the top 4" of the ground, loosening the soil, eating roots, and thinning the thatch. This activity begins in early to mid-August when the disturbed grass may wilt or die if conditions are dry. Damage is minimal when grub density is low or if the grass receives plenty of moisture. As the grubs grow, tunnels become vertical and deeper with turf damage becoming more severe. Grubs keep tunnels to the surface open by pushing little mounds of loose soil to the surface. The resulting mounds appear similar to earthworm castings. To determine that a mound was made by a green June beetle grub, wipe the mound away and feel for a hole in the ground about as wide as your finger. Earthworm holes rarely exceed the diameter of a pencil. The soil mound will reappear the next day. Fecal pellets about as big as mouse or small rat droppings may also be present on the soil surface near the holes. Fresh mounding activity is especially visible after a heavy rain. The mounds and holes are visible by mid-August, but the damage becomes more pronounced in the following months as the grubs continue to grow. The grubs do feed on some roots, but the major damage to the turf results from the upheaval of the acidic subsoil, dislodging of turfgrass roots from the soil, and subsequent weed problems.

The large green June beetle grubs come to the surface at night to feed or graze on the turf; individuals may migrate long distances (20'-30' per night). Grubs may also be found in the twilight hours and on overcast days. Their trails through the dew can frequently be seen on golf course greens.

Besides the direct damage, these grubs cause some indirect problems. The mounds and holes disfigure turf while the tunneling kills the grass. Drought-stressed turf mowed very short succumbs easily to this damage. As a consequence, spaces open up as the grass dies which enables weeds to establish. The tunneling and excavation of subsoil brings acidic soil to the surface and this changes the microhabitat that favors grass and broadleaf weed species. Turf managers using reel mowers have complained that the loose soil and grit from the mounds accumulates on the machinery and dulls the cutter blades, especially when the dew is still on the grass. Additionally, predators such as small mammals and birds damage turf as they dig for the grubs.

To date no thresholds are available for landscape turf or lawns. Treatments are recommended on perennial ryegrass/bentgrass golf course fairways when grub-counts exceed 5 per ft^2. Damage thresholds for Kentucky bluegrass and K31 tall fescue based on field observation are slightly higher at 6-7 grubs per ft^2. The K31 tall fescue variety with its broad leaf blades tends to hide damage better than the thin leaf blade species. Kentucky bluegrass will quickly recover with new growth from the underground stems.

To prevent damage to turf, apply controls to grub stages before many mounds become evident. We recommend an action threshold of five third instar larvae per square foot. Damage cycles historically run for 3-6 years then subside. During these outbreaks, damage may be expected if high populations of grubs were present the previous year and insecticide control was inadequate. An increase in the number of adults over the previous year's observations is also a reason to expect damaging populations of grubs.

NON-CHEMICAL CONTROL OF GREEN JUNE BEETLES

Biological Control

To date there are no effective commercial biological agents available to control this grub. The most common parasite is a type of digger wasp, *Scolia dubia* Say. This beneficial wasp enters the grub tunnel, stings the grub, then lays an egg on the paralyzed grub. The resulting larva feeds in the grub, eventually killing it. In the mid-Atlantic outbreaks during the 1980s, several golf course superintendents noticed an increasing number of digger wasps flying around the course about the time populations subsided. Unfortunately, even though these wasps help reduce the grub population, many people are afraid of being stung and consider them a nuisance. These wasps are not aggressive and need to be forced to sting a human.

Milky disease products effective against Japanese beetle do not control green June beetle grubs nor do any *B.t.* products.

Cultural Control

Some turfgrasses recover from damage once stress factors are removed. For example, species having stolons and rhizomes may repair the damage once the grub population is controlled. Also, the damage resulting from the grub tunneling is less severe when the turf receives sufficient moisture, fertilizer, and lime. Overseeding in the fall is critical in preventing weed encroachment the following season.

It is helpful to remember that grass cut at a greater height (2 1/2"-3") is less stressed and therefore the damage is less visible. Also, grass species such as K31 tall fescue with the broader leaf blades hide damage better than the fine-bladed grasses such as perennial ryegrass, bentgrass, or fine fescue.

CHEMICAL CONTROL OF GREEN JUNE BEETLES

Insecticides are effective on all grub stages and applications may be warranted anytime between August and November, as long as damaging numbers remain active. Spring applications of chemicals are not generally recommended since the grubs are active only for a few weeks and many may have pupated by the time damage becomes obvious. Once the grubs reach the third instar in August or September, they migrate freely and can easily move from an infested area to an adjacent area. To protect golf course greens, treat the greens, collars, and a few yards beyond the collars. The insecticides normally used to control sod webworms, cutworms, and armyworms on the greens will generally suppress migrating grubs. If fairways are treated, the rough areas should be spot-treated where there are high grub populations. The risk of high grub populations is generally correlated with areas where the adult beetle populations were most concentrated.

Most insecticides labeled for Japanese beetle grubs will effectively control Green June beetle grubs. Even insecticides that do not penetrate the thatch layer can work because Green June beetle grubs come up to the surface and become exposed. To control the early instars before the migration phase, application of insecticides must be followed immediately by irrigation with 1/2" of water, or timed with rainfall. Summer applications of isofenphos have not effectively controlled this species.

A word of caution is appropriate concerning insecticide treatment. After treatment, the grubs come to the surface within 12 hours and die, causing a foul odor as they decay. Turf managers should consider the possibility that these poisoned grubs may be eaten by other animals or domestic pets.

Finally, treated areas should be monitored carefully because migrating grubs may reinfest an area once the insecticide has broken down, making retreatment necessary.

ARMYWORMS

IDENTIFICATION AND BIOLOGY

True armyworm (*Pseudaletia unipuncta* [Haworth]) adults are buff to sand-colored, with a wing spread of about 1/2". Each forewing has a central white dot, and each hindwing has a dark margin. Fully-grown larvae, the turf-damaging stage, are nearly hairless, smooth, green to brown, have one dark stripe along each side, and a broad dark stripe along the upper surface. The top stripe may have a light, thin, broken line along its center. These stripes run the entire length of the body. The head of the larva is light brown with a green tinge and dark brown mottling.

Fall armyworm (*Spodoptera frigiperda* [Smith]) adults resemble those of true armyworms in form, but have dark gray forewings mottled with light and dark spots, and grayish-white hindwings. Larvae are gray to yellow-green, and have stripes similar to those of *P. unipuncta*. Each larva has a prominent, white, inverted Y-shaped marking on the front of the head. Long hairs arise from black tubercles along the body.

The true armyworm occurs throughout the United States east of the Rocky Mountains, as well as in New Mexico, Arizona, and California. The fall armyworm occurs throughout the United States in the warmer months, but is found all year in the southern states. This species overwinters in the Gulf Coast states and Florida and continuously migrates north during the spring and early summer.

The true armyworm attacks grasses, small grains, corn, alfalfa, sugarbeets, clover, and tobacco. Within turf and pastures, true armyworms inhabit the thatch layer. Under extremely dry conditions they will seek harborage inside soil cracks and under ground litter. The fall armyworm attacks grasses, corn, cotton, alfalfa, clover, peanuts, tobacco, and many garden plants. The microhabitats in turf, pastures, and meadows are very similar to those selected by the true armyworm.

The true armyworm passes the winter as a partially-grown larva in the soil or under debris in grassy areas. Activity and growth are continuous except during very cold weather. Larvae which successfully overwinter feed during the following spring. When fully grown, they stop feeding for four days, then pupate for 15-20 days. Adults emerge in May and June. Mating takes place at night, especially during the fifth hour after sunset (Pfadt, 1978); multiple matings usually occur. Females feed for 7-10 days on honeydew, nectar, or decaying fruit before laying eggs. Eggs are laid at night in clusters of 25-134 on grass or grain leaves. A single female may live as an adult for 17 days and produce up to 2000 eggs. Eggs hatch in 6-10 days. Young caterpillars begin feeding on leaves, especially at night or during cloudy weather. They usually hide in the thatch during daylight hours. Six larval instars are passed in 3-4 weeks; the last instar consumes 80% of the foliage eaten during the insect's lifespan. Full-grown larvae pupate in flimsy silk cocoon under litter or in earth cells 2"-3" below the soil surface. Following pupation in August or September, the emerging adults mate and lay eggs. Larvae develop partially before winter. The number of generations produced each year increases as latitude decreases; three to four are produced in the central states, while five or more generations are produced in the south. Outbreaks are most common after cold wet spring weather. In seasons of unusual abundance, larvae may crawl in large groups from one food source to another (hence their common name). Armyworms cannot survive exposure to temperatures above 88^{o}F. The fall armyworm life cycle is similar to that of the true armyworm. Although primarily a pest in the South, fall armyworm adults migrate northward each year and have reached pest status as far north as Minneapolis (Niemczyk 1981). Successful overwintering occurs only in the South.

MONITORING AND THRESHOLDS FOR ARMYWORMS

Defoliation damage is nearly identical to sod webworm injury. The only difference is that it proceeds at a faster rate because of the large size of the caterpillars. Synchronous egg-laying and subsequent population growth also contribute to the increased defoliation rate. Both species may be active throughout the growing season and outbreaks may coincide with sod webworm activity.

True armyworm treatment may be triggered when May larval populations reach one per ft^2. K31 and other fescues can suffer severe late-summer damage if fall armyworm larval populations reach one per ft^2. Other grasses may be more tolerant.

Methods used to sample sod webworms larvae will also work to detect armyworms. Because these two moth species are important agricultural pests, most states monitor the seasonal flight activity with blacklight traps. Your regional National Park Service Integrated Pest Management coordinator should have more information about monitoring programs in your state. Outbreak predictions for corn and other agricultural crops can also be a strong indicator that problems may occur in landscape turf situations.

NON-CHEMICAL CONTROL OF ARMYWORMS

Both of these species are susceptible to a wide variety of pathogens that occasionally become epizootic during major outbreaks. Several *B.t.* products will control the true armyworm but the fall armyworm requires special strains that are not presently labeled for turfgrass use.

True armyworm larvae may be effectively controlled by the parasite tachinid fly *Winthemia quadripustulata*. Other insect parasites include *Telenomus minimus* (an egg parasite); the braconid wasps *Apanteles laeviceps*, *A. marginiventris*, and *A. militaris*; ground beetles; sphecid wasps; birds; toads; domestic fowl; and small mammals such as skunks.

Parasites of fall armyworm eggs and larvae include the ichneumon wasp *Ophion bilineatus*; the braconid wasps *Chelonus texanus*, *Meterous laphygmae*, and *Apanteles* spp.; *Trichogramma minutum*; *Euplectrus* wasps; and the tachinid flies *Winthemia quadripustulata* and *W. rufopicta*. Predators include ground beetles, birds, and many small mammals.

Cultural Control

Since armyworm damage is similar to scalping the turf with a lawnmower, watering and fertilizing will quickly stimulate regrowth. Turf varieties with high levels of fungal endophytes are highly resistant to both species.

CHEMICAL CONTROL OF ARMYWORMS

Both species prefer to feed at night, so insecticides should be applied in the evening. Most insecticides labeled for sod webworm control will also control both armyworm species. Consult your National Park Service regional Integrated Pest Management coordinator for information on insecticide selection and timing in your area.

SOD WEBWORMS

IDENTIFICATION AND BIOLOGY

Most of the key turf pest sod webworm species are moths in the genera *Crambus, Pediasia, Parapediasia*, and *Fissicambus*. These moths are small, beige to gray-white, some with distinct color bands with a wingspread of 3/4". The head has a snoutlike projection at the front. Wings are folded and wrapped partly around the body when the moth is at rest. The larvae are caterpillars which may be greenish, brown, beige, white, yellow, or gray, depending on the species. They are 3/4" long when mature and usually have dark, circular spots scattered over the body. They spin threads of silk as they move, webbing leaves and soil particles together, and often form silk tubes in which they live. Consult Tashiro (1987) for additional information on the most common species encountered in turfgrass. The single most important species, *Parapediasia teterrella*, the bluegrass webworm, is detailed as an example for the typical webworm life history. This native species is very common in areas where Kentucky bluegrass is indigenous in the eastern half of the United States.

All species live in the thatch layer in grasslands, lawns, golf courses, rights-of-way, cultivated pastures, and sod farms. Although Kentucky bluegrass and fine fescue blends appear to be the most seriously affected, most cool season cultivated turfgrass species host one or more species. Evidently, slight variations in microhabitat can play a critical part in delineating damage. In Maryland, it was observed that severe outbreaks (10-30 larvae per ft^2) of the larger sod webworm and the bluegrass webworm are restricted to turf areas receiving nearly a full day of sunlight. Areas adjacent to fences, trees, shrubs, or buildings that provide partial shade usually host nondamaging populations. These shaded areas may also be less stressed during the drought periods that exacerbate webworm injury.

Depending on the species, length of growing season, and geographic location, the number of generations per year can vary from one to four (or more). The bluegrass webworm, for example, has two generations per year and overwinters as nearly mature larva within a silk-lined tunnel in the soil or thatch. The larvae feed during the evening and pupate within a cocoon at the end of its silk tube. In Maryland, adults emerge in late May and early June, mate, and lay eggs for several days. A second flight occurs in July and August with the mature larvae overwintering. Adults of all species are active at night. During the daylight hours, they rest in the thatch or on broadleaf plants near the turf. Eggs are nonadhesive and randomly drop into the thatch. Generation times are 4-10 weeks depending on the temperature.

MONITORING AND THRESHOLDS FOR SOD WEBWORMS

The sod webworm complex represents over 20 species in the United States, but the leaf feeding damage and the construction of the silken tunnels appear to be behaviors common to all species. Generally, the high-risk period for damage is from mid-July to the end of September, when many of the cool season grasses go dormant. The presence of one to two larger species caterpillars, or three to four small species in Kentucky bluegrass or fine fescue turf, is sufficient to cause defoliation during late summer. The damage is described as a browning of the turf; in reality the armyworms consume the green foliage during the night and the dead thatch layer appears as browning after heavy feeding. The damage caused is equivalent to a severe scalping of the turf like that caused by dull mower blades. Most bluegrass will partially recover when fertilized and watered to break summer dormancy. Fine fescue and perennial rye frequently die from the complete defoliation.

Larval damage can be expected 10-14 days after observations of heavy adult flight activity. Adults are highly attracted to blacklights, so this method can help indicate areas at risk for damage. High trap counts may not always correspond to high damage levels because of insect predation on webworm eggs and larvae.

Larval populations can be sampled using a visual thatch inspection or an irritant flush. Because the small first and second instar larvae are very difficult to find in the dense thatch, visual inspection tends to underestimate populations. The pyrethrin or soap flush, if too concentrated, may also underestimate by population numbers by killing the larvae immediately and allowing only the mature larvae to exit the thatch.

Excessive bird feeding activity in the turf may also indicate the presence or overabundance of mature larvae.

NON-CHEMICAL CONTROL OF SOD WEBWORM

Biological Control

Webworms support a wide range of native predators and parasites. The major predator of the eggs and young larvae are ants, predator, mites, and big-eyed bugs. Older larvae fall prey to birds, ground beetles, parasitic flies, and wasps. Several pupal parasites are well established in the northeast and together all these agents usually keep webworm populations below aesthetic thresholds. Frequent use of insecticides kill these beneficial species and their populations may require one to two years to recover.

Several naturally-occurring *Beauveria* fungal and *Nosema* and *Phelohania* microsporida diseases have been recovered from field-collected larvae. However, the Yimpact of these and other pathogens is poorly understood and probably greatly underestimated.

To date several commercial *B.t.* products and various species of parasitic nematodes provide good larval control. Both products are limited, however, because of the restricted labeled uses and the costs for multiple applications required to control multiple webworm generations throughout the summer.

Cultural Control

The warm-season turf species such as zoysia and bermuda appear to be resistant to webworm feeding. Tall fescue varieties are intermediate with Kentucky bluegrass, fine fescue, hard fescue, and perennial rye species, varying from highly susceptible to moderately resistant. Unfortunately, the selection for turfgrass insect resistance appears less important and more difficult to obtain in breeding programs than other characteristics. Major success has been accomplished in selecting for drought and disease resistance, Ph and fertility tolerances, green color, turf density, and persistence.

Fortunately a newly-discovered enhanced resistance due to a fungal endophyte offers plant breeders a combination of a high level of insect and drought resistance, plus some limited disease tolerance. Endophytes are fungi or bacteria that live inside a plant but do not cause disease. Instead, they enhance a grass's ability to survive drought, disease, and insect attack. At this time a limited number of varieties of tall fescue, perennial rye, and fine fescue possess high levels of protective endophyte. Varieties with endophyte infection levels of 70% or above are recommended.

The following variety tables are the most recent (1991) information on levels of endophyte:

Table 1. Endophyte Levels for Perennial Ryegrass

% Endopyte content in seed*				
Variety	**Hi**	**Mod. Hi**	**Mod. Lo**	**Lo**
Yorktown	97			
Palmer II	97			
Gen-90	97			
Express	97			
Advent	97			
Seville	06			
Dandy	96			
Duet	93			
ManhattanII	93			
PreludeII	93			
Repell II	92			
Assure	92			
Pleasure	92			
Target	92			
Riviera	91			
Gettysburg	91			
Pennant	91			
Legacy	90			
4 Del. Dwarf	90			
Pinnacle	90			
Repell	89			
SR 4200	89			
Commander	88			
Regal	88			
Saturn	85			
Competitor		71		
Accolade		70		

Equal		68		
Calypso		66		
Citation II			59	
Stallion			58	
Caliente			54	
Premier			50	
Entrar			47	
Prestige			43	
Derby Supreme			38	
Lindsay			37	
Charger			34	
Envy			30	
Rodeo II			27	
Essence				20
Fiesta II				15
Cowboy II				12
Danilo				06
Ovation				05
Loretta				04
Allegro				01
Gator				01
Danano				01
Pennfine				01
(Zero endophyte in other varieties.)				

Table 2. Endophyte Levels for Fine Fescue

% Endopyte content in seed*				
Variety	**Hi**	**Mod. Hi**	**Mod. Lo**	**Lo**
Jamestown II	100			
Reliant	100			
Warwick	96			
Southport	94			
SR-5000	92			
SR-3000		64		
Rainbow		63		
Valda		47		
Bridgeport		26		
(Zero endophyte in other varieties.)				

Table 3. Endophyte Levels for Tall Fescue

% Endopyte content in seed*				
Variety	**Hi**	**Mod. Hi**	**Mod. Lo**	**Lo**
Titan	98			
Shenandoah	86			
Mesa		70		
Tribute		58		
Aguara		50		
Arid			48	
Normark 99			42	
Rebel Jr.			37	
Trident				28
Rebel II				28
Winchester				24
Taurus				18
Apache				18
Finelawn I				16
Sundance				14
Thoroughbred				14
Murietta				14
Bonanza				12
Chieftain				06
Hubbard 87				04
Finelawn 5GL				02
(Zero endophyte in other varieties.)				

*NOTE: The data from tables 1,2, and 3 are from Rutgers University and were obtained from seed lots submitted to the National Turfgrass Evaluation Program. Seed lots may contain lower percentages of seeds with viable endophytes because of loss of viability during seed storage. (Source: Dr. Richard Hurley.)

Endophyte-infected grasses control insects in two ways; they repell the insects from feeding and poison them if they do feed. Research at the University of Maryland indicates that the first and fourth instar larvae are highly sensitive but the older larvae are slow to respond to the endophyte toxins. The young larvae die rapidly, one to six days after feeding, whereas older larvae stop feeding and die after five to six days.

Unfortunately no endophytes have been discovered in Kentucky bluegrass, but susceptible varieties with strong rhizome systems are more likely to recover than susceptible clump types. Irrigation and fertilizer applications in the fall will break the bluegrass dormancy and stimulate regrowth. However, complete recovery may require two years and the weakened turf may become more susceptible to summer diseases.

CHEMICAL CONTROL OF SOD WEBWORMS

All species of webworms are easily controlled with any of the presently registered insecticides. Control is usually accomplished within 12-24 hours after treatment. Consult your National Park

Service regional Integrated Pest Management coordinator for more information on use of pesticides in your area.

BLUEGRASS BILLBUG AND HUNTING BILLBUG

IDENTIFICATION AND BIOLOGY

Eight important species of billbugs attack turfgrass. Adults are gray, black, or brown, usually 1/4"-1/2" in length depending on species. These ground colors are often obscured by soil that adheres to pits, punctures, and grooves on the cuticle. Once cleaned, the surface sculptures on the pronotum and wing covers can be used to separate the species. Billbugs are weevils with a unique characteristic snout. These eight billbug species all have the antennae attached at the base of the snout nearest the eyes. The other major grass-feeding weevils have the antennae attached near the tip of the snout.

The two most common species are the bluegrass billbug, *Spenophous parvulus*, and the hunting billbug, *Spenophous venatus*. The *Sphenophous* larva is white and legless with a brown head capsule. The larvae of the various species are nearly identical and almost impossible to identify. Eggs are sausage-shaped, clear to creamy white. The female inserts these eggs individually into the stem or leaf sheath at the base of the plant.

The drawings and key by Shetlar (1982) should help identify the adults. Specimens should be washed in order to clearly see the diagnostic picture patterns; billbug adults are frequently encrusted with mud. They occur throughout the United States.

The bluegrass billbug is a potential key pest wherever Kentucky bluegrass is grown. This species also attacks perennial ryegrass, fescues, and timothy. The hunting billbug is most serious in zoysiagrass and bermudagrass, although it will feed on other species as well.

Turf with accumulated thatch offers the adult good overwintering harborage and protection from predators. In home landscapes, accumulations of leaves, pine needles, and bark mulches adjacent to turfgrass also provide good protection. Landscape sites that conserve heat during the early spring, such as sidewalk, driveway, asphalt pathway, and concrete or brick walls, encourage females to congregate and deposit higher numbers of eggs in the adjacent turf. Other turf sites in full sunlight are preferred over shaded areas for the same reasons.

Both species have identical one-year life cycles. The adult overwinters, feeds and disperses for a short period, and then begins egg-laying in May and early June in the mid- Atlantic states and Ohio. The adult female chews a small hole in the grass stem or leaf sheath and deposits an egg.

The first instar larva stage feeds inside and hollows the stem, resulting in an accumulation of a light tan powder or fine sawdust-like material inside the stem. The same material is evident in the crown area where the growing larva exits into the soil. The presence of this sawdust-like material helps separate billbug damage from disease injury. The older larvae feeds on roots at the thatch-soil interface.

A partial overwintering second generation will occur in the southern states. In warmer climates older larvae can be found in soil throughout the year. This results from a prolonged egg-laying period in the spring and late summer.

Pupation occurs in mid-July to early August and with the adult emerging between August to September to disperse for winter. Adult populations typically peak in April to late May and again in September to October. Peak larval damage is late June to July.

MONITORING AND THRESHOLDS FOR BILLBUGS

Both species of billbugs damage turfgrass in two ways. The early damage occurs in late June into mid-July. At this time the larva tunnels into the stem and crown causing the stems to brown and die. A good diagnostic sign of this kind of billbug injury is the compacted frass found inside the dead stems. The second type of damage occurs when older larvae feed on roots. These larvae are often intermixed with white grub larvae in the soil.

The collection of six to eight adults in a five-minute search constitutes a moderate infestation. These observations are made on sidewalks, driveways, and patio areas around buildings. Adult billbugs rarely fly, usually walking from lawn to lawn. This sampling method is limited because of the daily variation in adult activity caused by differences in weather, site microclimate, sampling time during the day, and landscape topography. Not all billbugs are active at one time and most are inactive on cool, rainy days. Monitoring done under these conditions will result in less accurate population estimates.

A preferable sampling method is the use of pitfall traps. This method continuously samples populations throughout the adult activity period from early April to late May when egg-laying begins, providing a more accurate population estimate. When trap counts range from two to five adults per day, moderate, spotty turf injury can be expected in two to three weeks. Severe losses can occur when adult counts exceed seven to ten per day over several days of sampling.

Ideally, pitfall traps should be used in combination with degree-day heat unit models and visual scouting. The degree-day models help predict insect activity based on temperature, and serve as a better estimate of peak activity periods during years with abnormal growing seasons.

NON-CHEMICAL CONTROL OF BILLBUGS

Biological Control

Naturally-occurring fungal diseases frequently kill adults during cool, prolonged, rainy periods during the spring and late fall. No fungal agents are registered in the United States. This may change within three to four years as overseas products are tested and labeled for the United States market.

Several commercial strains of entomophathogenic nematodes look promising for billbug control.

Steinernema glasseri, *S. feltae*, and *Heterorhabtis* spp. nematodes and other experimental strains are all effective, but no commercial products are available to date. *S. caropocapsae* species are not recommended for billbug control.

Cultural Control

Watering and fertilizing to stimulate regrowth of Kentucky bluegrass is an important technique for recovery from billbug injury. Varieties of grasses with extensive rhizome and underground stem systems are better able to quickly recover from billbug damage. Tall fescue and perennial rye grasses appear to better tolerate billbug damage, but once severely damaged they may not recover. Avoid using Kentucky bluegrass cultivars in problem situations that historically host high billbug populations. Some resistant varieties are available but these may have limited geographic usefulness because of other agronomic factors. This limitation may be partially overcome if fungal endophytes can be bred into these susceptible varieties to make them more widely adaptable.

Endophyte-infested tall fescue and perennial ryegrasses are excellent sources of resistance and highly recommended for use in high-risk billbug areas.

CHEMICAL CONTROL OF BILLBUGS

An adult control program offers the best option when billbugs become habitual problems. Two behavior characteristics support this choice. Since the adult female rarely flies, migration into uninfested turf areas each spring is slow. Once established, they require a pre-oviposition period of two to three weeks before egg-laying. Thus an April to mid-May application of insecticide will kill the adult females before they lay eggs. Since there is only one generation per year, reinfestations may not occur again for two to three years after treatment because of the limited migration mentioned above. Billbug grub control is generally less satisfactory than the adult treatments.

MOLE CRICKETS

IDENTIFICATION AND BIOLOGY

Four species of mole crickets are commonly associated with turfgrass and pastures in eastern half of the United States. Two species, the southern mole cricket (*Scapteriscus acletus*) and the tawny mole cricket (*S. vicinus*), are major turf pests in the southeastern United States. The shortwinged mole cricket (*S. abbreviatus*) and the northern mole cricket (*Neocurtilla hexadectyla*) are less important as pests. Adults are 1/2"-2" long, and may be grayish-brown to brown in color, with enlarged heads, beadlike eyes, short antennae, and robust, spadelike front legs with large spikes used for digging. Forewings are usually half the body length, while the hindwings may extend beyond the tip of the abdomen when folded except in the short-winged species. Mole cricket nymphs resemble adults but are smaller and wingless. Eggs are deposited in clusters in the soil. The northern mole cricket has four claws on the front pair of legs, while the other species have only two.

The shortwinged, southern, and tawny mole crickets occur in the south- Atlantic and Gulf Coast regions from Virginia to Texas. The shortwinged species is restricted to coastal Florida and Georgia. The southern mole cricket is the most widespread of the three. The northern species is found throughout the eastern half of the United States.

The southern and tawny mole crickets have very similar life cycles, habits, and destructive behavior. Nymphs and adults migrate deep into the soil during cool weather and pass the winter in the soil. In spring and early summer, females construct cells in the soil in which they lay about 35 eggs are laid in each cell. These hatch in 10-40 days, depending on ambient temperatures. Nymphs grow rapidly, most becoming adults before fall. Those emerging from eggs laid late in the season will become adults in the spring of the following year.

Nymphs and adults of both the southern and tawny species feed on various warm- season grasses. Bermuda and bahia grass appear to be the major hosts. Mole crickets also feed on garden vegetables, tobacco, peanuts, and strawberries. Underground stems, roots, tubers, and fruits touching the ground may also be damaged.

Mole crickets are rarely found in heavy soils, preferring sandy to sandy loam soils. Regardless of texture, moderate to high soil moisture is a prerequisite in maintaining the open tunnel structures. Nightly tunneling ranges from 5'-20'. Both nymphs and adults hide in this tunnel system by day and emerge at night to feed on plants, roots, organic matter, and other arthropods. They can be cannibalistic, particularly the southern species.

MONITORING AND THRESHOLDS FOR MOLE CRICKETS

Immatures and adults burrow in loose soil, feed on grass roots, and cause the turf to dry out. Damage is usually localized in irregular areas and can be severe in newly- planted turf. Infested areas may feel soft underfoot due to the tunnels under the thatch. Suspect turf can be visually inspected for the presence of entrance holes (1/2"-1" in diameter) in the soil or thatch layer. A quantitative estimate of mole cricket populations is obtained using the soap flush technique. A solution of 1-2 ounces of liquid dishwashing soap in 2 gallons of water is sprinkled over a marked 4 ft2 area of turf. The soap irritates the insects, driving them to the surface. All mole crickets coming to the surface in the three-minute period following treatment are counted and the total is divided by four to convert to number per square foot (Short et al. 1982). Short et al. (1982) determined the economic threshold for this pest at 2 per 0.1 m^2 for bahia grass lawns in Orlando, Florida.

Other thresholds are based on a 4 ft sample area using either a soap or pyrethrin irritant flush. Insecticides are usually recommended after the emergence of 7 mole crickets within 3 minutes. Several researchers noted examples of severe damage to golf tees caused by mole cricket populations greater than 1 per square foot.

Another source of aesthetic damage is the mound of soil that accumulates at the tunnel entrance.

These small mounds produced by the tunneling nymphs and adults will cause problems on golf course greens and tees, regardless of population levels. Their presence also interferes with play and mowing activities.

NON-CHEMICAL CONTROL OF MOLE CRICKETS

Biological Control

Several researchers report that the green disease (metch), which is caused by the fungi *Metarrhizium anisopliae*, and the red disease, which is caused by the fungi *Sorosporella uvella* (Kass), will infect mole crickets. However these naturally-occurring fungal diseases are not consistent from year to year. Several commercial products with the *Metarrhizium* fungi are available outside the United States, but similar products are expected to be registered here by 1994.

The most promising biological agent to date is the parasitic nematode *Steinernema scapterisa*. Preliminary research by Parkman and Frank at the University of Florida demonstrated that sound traps using two Mans-type emitters, each set for a single species, can be used to attract and expose mole crickets to this nematode. Early studies also determined that this nematode could be released once and persist for several years in pastures. Commercial products with this unique mole cricket nematode should be available after 1993.

Natural occurring parasitism and predation is limited, but fire ants, ground beetles, spiders, and mole crickets themselves do help regulate populations. Two imported parasites have been released in Florida and show some promise. The sphecid wasp *Larra bicolor* (americana), which is found in southern Florida, seeks out the mole cricket in the tunnel, paralyzes it, and lays an egg on the thorax. In about two weeks the developing wasp larvae kills its host. The other imported parasite is a tachinid fly called *Ormia depleta*. This fly is attracted to the sound produced by the mole crickets. After finding the mole cricket, the fly lays an egg on the host; the developing maggot stage then kills the host.

Cultural Control

The combination of fertilizing, irrigating, and rolling the turf in the spring will lessen the spring tunneling damage. The rolling helps prevent the root system from drying out in light sandy soils.

CHEMICAL CONTROL OF MOLE CRICKETS

Because mole cricket migratory flights occur twice a year and both adults and nymph tunnel extensively, the risk of continuous reinfestations is high. Therefore, two insecticide treatments may be required. Contact insecticides are most effective against the newly-hatched crickets. With late-season outbreaks or with early spring tunneling, short-residual insecticides are recommended. Insecticide baits also work well against the nymphs, but are relatively ineffective against adults in fall. Regardless of the insecticide strategy selected, adults are very difficult to control. Consult your National Park Service regional Integrated Pest Management coordinator for more information on chemical control of this pest in your area.

CHINCH BUGS

IDENTIFICATION AND BIOLOGY

Two species of chinch bugs are of primary concern in turf; the southern chinch bug (*Blissus insularis*), a pest of warm-season turfgrasses, and the hairy chinch bug (*Blissus leucopterus hirtus*), a pest of cool-adapted turfgrasses. Adults are black, about 1/5" long, with white wings folded over the back. Each wing is marked with a dark triangle on the outer margin. As many as 80% of these individuals may have short wings (about 1/2 the length of the body), while the wings of others extend to the tip of the abdomen. The legs and the bases of the antennae are red. Juveniles (nymphs) resemble adults, but are wingless; the youngest nymphs are bright red with a single white band across the middle of the body, but darken as they mature. Eggs are elongate and about 1/15" long and usually inserted into the leaf sheath or crown area.

Chinch bugs occur throughout the United States. The southern chinch bug is a pest in the Gulf states, Georgia, South Carolina, and North Carolina. The hairy chinch bug is a pest of the more northern states.

Chinch bugs overwinter as adults, hiding in tufts of bunching grasses, under litter or leaves at woodland borders, under hedges, in fence rows, or in crop stubble. Winter inactivity may be broken by abnormally warm weather. Generally, migration from winter sites begins after one or two sunny days with temperatures of 70 ^{o}F or more. Females lay eggs in leaf sheaths, in the crown-area roots, or on the soil near host plants. Fifteen to twenty eggs per day are deposited in two to three weeks. Eggs hatch in one to two weeks and nymphs begin sucking juices from host plants. The bugs pass through five instars in 30-90 days before reaching adulthood. The eggs produced by this generation of females become the second generation of adults in late summer and early fall. From August to October, these adults gradually migrate to their overwintering sites. In the south and southwest, three or more generations may be produced before hibernation.

MONITORING AND THRESHOLDS FOR CHINCH BUGS

The chinch bug prefers the following grasses listed in order of importance:

Hairy chinch bugSouthern chinch bug
BentgrassSt. Augustine
Fine fescues
Perennial ryes
Kentucky bluegrass
Zoysia
Tall fescues

Chinch bugs damage grass plants by inserting their hollow beaks into the stems, sucking the plant juices, and injecting chemicals into the plant which clog the vascular system. The area around the feeding puncture usually turns yellow. Damage appears as patches of dead or gradually yellowing grass, especially where heat is radiated into the grass from sidewalks or roadways. Once the grass turns brown, the turf will not recover.

Reseeding or renovation is usually necessary after moderate damage. Usually 15-20 chinch bugs per square foot will require treatment. Chinch bugs prefer warm, sunny, dry locations. Adults rarely fly in the mid-Atlantic region. However, in Canada, where the short-winged form is limited in numbers, adults frequently fly from lawn to lawn. The best way to monitor chinch bug activity is by flotation sampling, which is described in the section on monitoring at the beginning of this module.

NON-CHEMICAL CONTROL OF CHINCH BUGS

Biological Control

Naturally occurring fungal diseases such as *Beauveria globcelifera* regularly control chinch bugs during cool, wet weather. Another regulating biological agent is the big-eyed bug, which can destroy entire populations; unfortunately, chinch bugs frequently cause serious damage before big-eyed bug populations peak. The wasp *Eumicrosoma beneficum* may parasitize up to half of the chinch bug eggs in favorable locations.

Commercial parasitic nematode products which contain the nematode *Steinernema carpocapsae* are effective against chinch bugs.

Resistant Turf Varieties

Several highly resistant varieties are now marketed, particularly for the southern species. The cooperative extension service can provide variety recommendations for your geographic area. Recently discovered fungal endophytes provide enhanced resistance. Chinch bugs both avoid turf plants with endophyte or die quickly after feeding. The naturally- resistant St. Augustine grasses and the endophyte-enhanced varieties offer the best long-term, environmentally-sound control choice available at this time.

Research at the University of Maryland has documented an immediate, rapid decline in mortality in young nymphs feeding on the endophyte-infested turf varieties. A similar but slower mortality rate was observed among the adult insects.

CHEMICAL CONTROL OF CHINCH BUGS

Resource managers usually control populations after major damage has occurred. To avoid this problem, areas with habitual problems should receive an insecticide application in April to mid-May that will control the overwintering females and subsequent generations during the summer. Reinfestation may occur from adjacent areas, but this process is slow and may require an additional year. This treatment must be made before egg-laying occurs in early to mid-May in the mid-Atlantic states. Consult your National Park Service regional Integrated Pest Management coordinator for more information on chemical controls in your area.

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Trade Magazines Related to Turf

Grounds Maintenance
P.O. Box 12901

Overland Park, KS 66282-2901
601-624-8503

Landscape Management
7500 Old Oak Blvd.
Cleveland, OH 44130
216-243-8100

Lawn and Landscape Maintenance
4012 Bridge Ave.
Cleveland, OH 44113
216-961-4130

Southern Turf Management
P.O. Box 1420
Clarksdale, MS 38614
601-624-8503

Turf
P.O. Box 391
50 Bay St.
St. Johnsbury, VT 05819
802-748-8908

Turfgrass Weeds

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for turfgrass weeds. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Weeds are usually described as any plants growing where they are not wanted. Any undesirable grass or broad-leafed plant species, from a small herbaceous plant to a woody shrub, vine, or tree, may be considered a weed if it is growing in a turf planting. Dicotyledonous (broad-leafed) plants are those that have two cotyledons in each seed. They are characterized by broad leaves and often have woody stems. Some species (e.g., sunflower) only become woody in old parts of stems and roots; these are referred to as semi- herbaceous dicots. Most weeds in turf have little or no woody tissue and are herbaceous (e.g., plantains, dandelion).

Grasses are members of the plant family Gramineae. All grasses are monocotyledonous, have long, narrow leaves with parallel veins, and fibrous root systems. Some grasses produce underground stems called rhizomes (e.g., Kentucky bluegrass, quackgrass) or above-ground runners called stolons (e.g., creeping bentgrass), while others (e.g., bermudagrass) produce both.

It is important to understand the distinction between monocots and dicots since turfgrass herbicides kill only dicots. (Desirable turf species are monocots.)

Why Weeds Invade Turf

Undesirable ("weed") plants will invade man-made environments such as turfgrass plantings wherever they are established. Weeds growing in turf are found where soil has been exposed or disturbed by compaction, planting activities, or maintenance activities such as sidewalk edging. For example, goosegrass and knotweed readily colonize heat-stressed and compacted soil sites along sidewalks or on athletic fields. They also occur where the turf is weakened by adverse environmental conditions (such as drought), thatch accumulation, diseases, or pests to the extent that the turf cannot compete for nutrients, water, or light with weed species. Exposure to de-icing salts, fertilizer or other chemical spills, and dog urine can also leave bare spots in which weeds will grow. Weeds are very common where the grass species being grown is not well-adapted to its environment. Many weed species possess efficient methods of seed dispersal such as wind dissemination of winged or hairy seeds or the ability to spread rapidly by rhizomes, runners, or tubers.

The description of each potential weed species is beyond the scope of this report. Contact the

Cooperative Extension Service of the agricultural university in your state or your Park Service regional Integrated Pest Management coordinator for specific information on the most important weeds in your region. A list of useful pictorial weed guides is included in the reference section.

Life Cycles

The life cycles of turf weeds can be grouped into the following major types.

Summer annual weeds. These are weeds that grow each spring or summer from seed. Examples include prostrate spurge, ragweed, large crabgrass, and goosegrass. They mature, produce seeds, and die in one growing season. Seeds generally overwinter before germinating the following spring. The majority of annual weeds are of this type. Some annuals, such as crabgrass, can root from leaf-stem junctions, forming dense colonies.

Winter annual weeds. These are weeds that germinate in the fall or late winter from seed, mature and produce seed during the following spring, and die in early summer (e.g., henbit, shepherdspurse, annual bluegrass). Seeds of most of these species are dormant during the spring.

Indeterminate annual weeds. These are weeds such as chickweed and annual bluegrass, that germinate and grow during most seasons in certain regions.

Biennial weeds. These weeds may germinate at any time during the growing season. Examples include wild carrot, bull thistle, and mullein. They usually produce a radial cluster (rosette) of leaves lying close to the soil during the first season. In the second year they produce flower stalks (using food stored from the first season's growth), produce seeds, and die.

Perennial weeds. These plants live for three or more years. Some species may not flower the first year, while others may produce mostly nonviable seeds. Many perennials (e.g., curly dock, dandelion, and common milkweed) spread primarily by producing seeds, while others (e.g., field bindweed, white clover, silverleaf nightshade, bentgrass, and quackgrass) spread both by seed and vegetatively. Vegetative spread can occur by rhizomes, stolons, tubers, or rooting of stem nodes that touch the soil.

The seasonal abundance of weeds is related to their specific life cycles. Summer annuals grow from spring until fall and are killed by low fall or winter temperatures. Winter annuals are present from fall to late spring, so they are usually not found during the summer. Biennials grow during the spring, summer, and fall of their first year, survive over the following winter, and flower during the next growing season. Therefore, some biennial stages are likely to be present at any time of the year. Perennials grow during each growing season. Their aboveground structures may die over the winter (e.g., Dallisgrass and yellow nutsedge) or may remain viable but dormant.

Knowledge of the life cycle of a particular weed species is an important part of its management. For example, mowing a patch of annual weeds to remove the flowers can prevent seed set. Refrain from cultivating areas where there are high populations of weeds which reproduce by rhizomes.

Impact of Weeds

The most obvious impact of weeds on turf areas is the competition with and replacement of turf by weed species. In the case of weeds that over grow an area and then die, such as crabgrass and knotweed, unsightly dead areas can be created. This often leads to the necessity for increased expenditures for turf renovation.

Toxicity to humans and animals is also a consideration. Some common weeds are poisonous if consumed (e.g., black nightshade, pokeweed, poison hemlock, and Johnsongrass); cause inflammation when touched (e.g., stinging nettle, and poison ivy, oak, and sumac); or cause allergic reactions (e.g., common ragweed and goldenrod). Visitor injury or annoyance can result from bees or wasps seeking nectar from some weeds. Furthermore, many weed plants or their seeds have spines, thorns, or burs which can create similar allergic effects.

Weedy areas provide habitat for desirable wildlife and beneficial insects but they can also harbor rodents and arthropods such as rats, ticks, mites, and fleas that might attack humans and domestic animals, or carry diseases that may infect humans and domestic animals. Weeds can also serve as secondary hosts for some fungal pathogens and insects that might attack desirable turfgrasses.

MONITORING AND THRESHOLDS FOR TURF WEEDS

Monitoring For Weeds

Monitoring for actively growing weeds should be done periodically throughout the growing season. Less frequent inspections should be made during winter and early spring to identify sites of soil disturbance or other adverse effects, which may give rise to future weed problems.

It is essential that all monitoring results be reported completely and accurately by site and date so that future surveys will cover the same areas. Recorded weed information allows the site manager to develop a weed history of an area. This will result in a more accurate prediction of future weed management needs.

Regular visual inspections of turfgrass areas should be conducted to look for actively growing weeds as well as newly germinated weed seedlings. Weeds are most likely to be found in areas where some type of disturbance has taken place, such as areas of soil compaction, areas of open soil, worn areas on athletic fields, or areas of soil moisture extremes. Weeds also are likely to grow in turf that has been stressed. This could be the result of being mowed too short, heavy thatch accumulation, competition from trees, or insect or disease attack. Turf can be stressed from extremes in soil pH or the accumulation of road salts along roadways as well. "Dog blight" from animal urine, fertilizer or pesticide spills or misuse, localized wet or dry spots, or accumulations of debris can create open areas where weed infestations begin. Edging along walkways may also open up areas of bare soil where weed seeds can germinate.

Certain weed species tend to be found in certain habitats, so monitoring for a particular weed should be based on a knowledge of its biology. For example, crabgrass is a spring annual that needs light to germinate. Therefore, crabgrass seedlings are most likely to be found in bare or thinning areas in the spring. If they are not found in areas such as this, it is unlikely that they will be found in a shaded area of denser turf. Also, since crabgrass is a spring annual, it may be a waste of time to look for seedlings in mid-summer. This would be an excellent time to look for mature plants, however, to identify seed sources for the following year. Late fall would be the best time to look for seedlings of winter annuals such as henbit or annual bluegrass.

Monitoring large areas of turf for weeds can be very time-consuming, so certain techniques should be employed to make the monitoring process more efficient. One commonly used technique in weed monitoring is to randomly walk 50' long lines of turf, identifying and counting all weeds which touch the line. Lines should be randomly placed in areas that represent all turf species, habitats (e.g., sun v. shade), or different use areas that may be present.

Action Thresholds for Turf Weeds

It is extremely difficult to set specific threshold population levels for weeds in turf since the problems caused by weeds are largely aesthetic, rather than medical or economic. Each park manager should establish threshold and action levels for his/her own area by maintaining records and scouting of weed populations in all turf areas. Action levels will be lower in high-use areas such as lawns around buildings and picnic or rest areas than they will be in large, unused turf areas or parking areas. Vigorous weed competitors such as crabgrass, white clover, and quackgrass should have a lower action threshold than other weeds.

NON-CHEMICAL CONTROL OF TURF WEEDS

Established Plantings

Employ sound cultural practices including regular soil testing, proper fertilization at the correct time, mowing at the correct height and frequency, and deep irrigation when needed. Frequent shallow watering discourages root growth and can encourage weed seed germination and some turfgrass diseases. Mow no shorter than 2.5" for cool-season grasses such as Kentucky bluegrass, tall fescue, fine-leaf fescues, and perennial ryegrass to prevent weakening of grass and encouragement of weed seed germination. Always remove debris and heavy thatch from turf and alleviate soil compaction through core aeration.

Renovate chronically poor turf sites to regionally adapted species and cultivars. Fall is the best time to do this for cool-season grasses, while spring is best for warm- season grasses.

Finally, develop a regular monitoring program for weeds and disturbed areas. Set a tolerance level for weeds and remove them mechanically or with proper application of a registered herbicide when that level is exceeded.

New Plantings

Plant turfgrass species and cultivars that are adapted to the growing area and, if possible, resistant to diseases and insects. Even though it is more expensive, use certified seed which is free of noxious weed seeds. Renovation and new plantings should always be done at the times of year that are most appropriate for the particular species; i.e., fall for cool-season grasses and late spring for warm-season grasses. When preparing the area for planting, allow weed seeds to germinate and then cultivate or apply a non-selective herbicide to kill young plants. Cultivation without use of a non-selective herbicide is generally not recommended for weeds that produce rhizomes, stolons, or bulblets because it breaks these structures into smaller pieces and may therefore result in dispersal rather than control of the weed.

During establishment of turf, inspect regularly for weeds. Mechanically remove weeds found in small populations or spot treat areas with a concentrated weed population with a registered herbicides. Always check the herbicide label for information on seedling tolerance.

Cultural Methods of Turf Weed Management

Turf management practices that increase the health, density, and general vigor of grass will discourage weeds through competition. It is essential to use turfgrasses that are adapted for the specific planting area (i.e., region, climate, light intensity) and type of use (e.g., heavy traffic). This will promote the best possible sod development. When turf is established or renovated, grass seed, sod, topsoil, and mulches that are free of weed seeds should be used.

Turf maintenance practices should stress proper fertilization and liming based on the results of soil tests. The amount of nitrogen and timing of its application are extremely important factors for maintaining turf density and discouraging weed encroachment. Consult your Park Service regional Integrated Pest Management coordinator for the correct information for your area. Deep watering (to wet soil to a depth of 5"-8") when grass begins to show signs of wilting will prevent the development of shallow root systems and weak turf, and will help to reduce weed, disease, and insect problems. Frequent, shallow watering encourages the germination of some weed seeds and should be avoided.

It is also important to remove leaves or other accumulated debris from turf, since this can smother or shade the grass, allowing weeds to grow in its place. Heavy thatch is reduced by a combination of core aerification, maintenance of soil Ph between 6.0 and 7.0, and use of balanced fertilizers with slow-release nitrogen. Thatch also can be avoided through the use of tall fescue or other bunch-type grasses (where adapted) and by avoiding excessive nitrogen fertilization.

Mechanical Methods of Turf Weed Management

Frequent mowing will prevent or reduce seed production in some weed species. A few weed species such as Johnsongrass or poison ivy can generally be removed from turf by scheduled mowing. Cool-season grasses should be cut no shorter than 2.5" in height to prevent weakening of the grass plant, and high mowing will promote a dense turf that can more effectively compete with weeds. Despite proper mowing, weeds may still become a problem in turfgrasses. Lower mowing is desirable for some grasses such as bermudagrass and zoysiagrass.

Biological Control of Turf Weeds

No biological control agents for weed control have been approved by APHIS for use in turf plantings.

CHEMICAL CONTROL OF TURF WEEDS

When chemical weed management is necessary, start by using spot applications only in areas where weeds are starting to dominate a stand. When needed, apply broadleaf herbicides primarily during fall or spring to mature turfgrass stands. Annual grass weeds are best controlled with preemergence herbicides applied in early spring, prior to weed seed germination. Keep in mind that proper herbicide selection and use can be complicated. Your regional Park Service Integrated Pest Management coordinator will have more information relevant to your particular situation.

An in-depth discussion on chemical weed control is beyond the scope of this module. However, information on selection of turf herbicides can be obtained from your regional Integrated Pest Management Coordinator, your local Cooperative Extension Service Agent, or your local agricultural state university turf specialist. Some basics of chemical weed control are as follows.

Weed control prior to establishment or renovation. In sites where extremely persistent, perennial weeds exist (e.g., quackgrass, bermudagrass, and red sorrel), a non-selective herbicide such as glyphosate should be applied twice on a 30-day interval. This is ideally done prior to tilling and a few weeks after tilling. The first application will kill existing weeds, while the second will kill weed seedlings which have germinated from seeds that were dormant in the soil. Being non-selective, glyphosate also will injure or kill desirable turfgrasses, flowers, and other herbaceous plants. Glyphosate, however, has no soil residual and treated areas can be reseeded within 24 hours of use.

When new areas of turf are being established, shallow cultivation will bring many buried weed seeds to the surface and allow them to germinate. This should be followed by an application of a non-selective herbicide such as glyphosate to control these weeds.

Perennial broadleaf weed control. These weeds are generally controlled or reduced to below threshold levels with a single fall or spring application of a selective herbicide. Do not use these herbicides unless there is sufficient soil moisture to support active growth of weeds. Air temperatures should range from 65 to 85F, and there should be no wind when these herbicides are applied. Liquid or sprayable herbicide formulations provide superior control than granular formulations. Carefully read and adhere to all information and directions outlined on the label.

Annual grass weed control. This group of weeds includes crabgrass, goosegrass, foxtails, sandburs, etc. These weeds are considered major weeds because they can effectively compete with grasses and can significantly reduce turf stand density in a single season. High mowing greatly retards annual grass weed populations, but when monitoring indicates increasing populations of annual grasses, an application of pre- emergent herbicide should be planned for the next spring. To be effective, these herbicides must be applied in early spring prior to weed seed germination and they must be watered-in by rain or irrigation within three to five days of application. Watering-in is critical and in the absence of irrigation these herbicides are best applied on a rainy day. Consult a turfgrass specialist or your county cooperative extension service for information about appropriate application times and herbicide selection in your area.

Postemergence annual grass weed control. The postemergence approach to annual grass weed control requires more knowledge and professional skill to be effective. Consult an extension turfgrass specialist in your region for more detailed information on herbicide selection and use.

An example of a turf weed management program which has been implemented by the National Capital Region of the Park Service follows.

Turfgrass Weed Management in the National Capitol Region

The National Park Service manages turfgrass in three of its management zones: developed, special use, and historic. Within each management zone the visual objective for turfgrass quality will depend on the management objectives established for the park and management resources available.

Generally, turfgrass can be classified into three classes: ornamental turfgrass, recreational turfgrass, and greenspace turfgrass. These classes will determine the type and level of maintenance each should receive.

Classes can be used in integrated pest management to determine thresholds and management strategies for specific pests. The following guidelines have been developed specifically for weed management in turfgrass to assist in determining where and when the use of herbicides would be appropriate.

The intent of these guidelines is to sustain the quality of turfgrass appropriate for specific sites, while minimizing dependency upon herbicides to achieve this quality. This can best be done by using a fully integrated turf management program which defines the site objectives, assures objective monitoring, and encourages cultural management practices.

TURFGRASS CLASSIFICATION

Class A. Ornamental turfgrass

Lawns classified as ornamental have the highest visual quality objective. Turfgrass must appear uniform in color and texture with weeds and bare spots unnoticeable to the general public. Ornamental lawns are exposed to minor foot traffic and receive the highest level of maintenance. Ornamental lawns provide the setting for memorials and other significant sites and features. Considering the intensive maintenance these areas require and that visual quality necessitates limited visitor use, managers should restrict the Class A designation to the minimum area necessary to achieve the visual management objective.

Class B. Recreational turfgrass

Turfgrass which provides the setting for certain passive recreational and athletic activities can be classified as recreational turfgrass. Class B areas would include small urban parks and some playing fields for organized sports, as well as turfgrass surrounding offices, parking lots, and other support facilities. Although such areas may have ornamental significance, the visual quality and level of maintenance is less demanding than ornamental turfgrass. Visitor use is common and some weed infestation is tolerable. The uniformity in color and texture is not as critical as in ornamental areas.

Class C. Greenspace turfgrass

Greenspace turfgrass encompasses large areas that receive minimal maintenance other than mowing. The aesthetic objective for the site is achieved simply by the presence of turf and not by its quality. Greenspaces would include large picnic and informal recreation areas, parkway medians, and roadsides.

Managers should classify all turfgrass within a park into these three classes. In some parks all three classes may occur contiguously. The classification scheme allows for managers to exercise their judgement and experience in classifying turfgrass. Classification, however, should be based on the parks statement for management and other mandates and factors establishing the visual objective for the site. In historic areas, class designation should be supported by documentation describing the appearance of the site during the period portrayed. All site classifications should be defined in the resource management plans and reviewed routinely to assure their proper designation.

ACTION THRESHOLDS

The application of herbicides in conjunction with other weed management tactics may be necessary to achieve the visual quality objective. Proposals to apply herbicides (Pesticide Proposal Form 10-21A) should be supported by the site classification and quantitative monitoring data showing that the action threshold for the class has been reached. The action threshold is expressed as the maximum percentage of weed cover allowable before herbicide applications can be made. Turfgrass should be monitored routinely and systematically, recording quantitative measures of weed cover.

Class A -- Ornamental Turfgrass

Spot treatment. Spot treatment may be considered when an action threshold of 15%- 29% weed cover is reached. Treatment must be limited to those areas that have reached the action threshold and can be sprayed with a backpack or other similar single nozzle, small capacity sprayer.

Recovery treatment. An action threshold of 30%-49% weed cover indicates a possible need for broadcast application of herbicides by backpack or tractor-mounted, multi-nozzle boom sprayer. Treatment must be preceded by a complete review of the turf management program to determine why the weed level reached the Recovery action threshold. The review will examine the level of use, compaction, turf variety, mowing height, moisture management, fertility, Ph, and other factors considered pertinent to maintain Class A turf.

Herbicide applications must be used in conjunction with other tactics to remedy the management deficiencies or site factors responsible for weed infestation. Recovery treatments will not generally be approved for consecutive years.

Renovation. When the action threshold reaches 50% or greater weed cover, complete renovation is warranted. Renovation will be preceded by broadcast application of glyphosate or other similar broad-spectrum, post-emergent, short-residual herbicide that kills all vegetation. The treatment must be preceded by review of the overall turf management program as described for recovery treatments. The site will be seeded or sodded as appropriate. All other management or site deficiencies determined in the review must be corrected.

Class B -- Recreational Turfgrass

Spot treatment. Not permitted in recreational turfgrass.

Recovery and renovation. As described for Class A turfgrass.

Class C -- Greenspace Turfgrass

Herbicide applications are not permitted on Class C turfgrass.

MONITORING

Monitoring is the most important component of any integrated pest management program. Only through routine monitoring can a manager be aware of changes in turf status. A proposal to apply herbicides must be supported by objective monitoring data demonstrating that the action threshold has been reached. Visual estimates are often inaccurate and therefore unacceptable as justification for a herbicide request. In addition to determining the percent weed cover, monitoring also determines the percent bare ground which is susceptible to weed invasion. Monitoring facilitates the identification of the weed species present. Species identification is critical in developing the appropriate management strategy. Post-treatment monitoring provides a measure of the efficacy of the overall management program.

The procedure and intensity in which monitoring is applied will depend on the size and uniformity of the area being examined. If the area of interest is not uniform in cover, it should be subdivided and monitoring performed in each subdivision. A minimum of five monitoring points should be used in any size area and no less than ten per acre. The more monitoring points used, the more definitive the assessment will be. However, monitoring must be simple, quantitative, and efficient. Before monitoring, there must be a consensus as to what constitutes a weed, since some species such as bermudagrass, annual blue grass, and clover may or may not be considered weeds depending on the site and its management.

Monitoring in small areas can be performed by randomly selecting monitoring points by the toss of a marker (rock, ball, etc.). The marker can be thrown at five random points in an "M" pattern spread over the expanse of the area to be examined. In areas of more than one acre, mark off a series of straight or zigzag lines and randomly select points along those lines. Measurements at the monitoring points can be made in a variety of ways. At each monitoring point a specific area of turf should be examined and the percentage of turf, weed, or bare ground determined. For example, a one foot square block of turf might be 33% covered by turf, 33% by weeds, and 33% bare. If possible, determine what weed species are present. This is important because the life cycle of a weed is a consideration in determining how to manage it. For example, it is not a good idea to cultivate an area with perennial weeds which produce rhizomes, since this will cause more plants to be produced. Winter annuals will set seed in early spring and could be mowed before this to prevent seed formation.

Safety

All herbicide applications must be made in the early morning or evening before or after visitors are present. Treatments must be made in a safe and responsible manner either by a licensed applicator or individual working under the "line of sight supervisor" of a licensed applicator. Treated areas must be posted.

Pesticide Approval Request (Form 10-21A)

In addition to the information already required, requests for herbicide use on turfgrass must include:

1) turfgrass classification, class A or B.

2) the percent weed infestation and weed species present,

3) the method used to determine the level of infestation, pre- and post-treatment, and

4) a statement of other management measures that will be taken.

Guideline Summary

Table 1 summarizes the herbicide treatment options based in turfgrass class and percent of weed cover. If herbicides are going to be part of your turf weed management strategy at a particular site, this will help you to decide the most appropriate way in which to ue them.

Table 1. Herbicide treatment options based on turfgrass class and weed cover.

% Weed Cover	Class A	Class B	Class C
0% to 14%	Cultural	Cultural	Cultural

15% to 29%	Spot	Cultural	Cultural
30% to 49%	Recovery	Recovery	Recovery
> 50%	Renovation	Renovation	Cultural

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Weeds of Developed and Historic Sites

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for weeds in historic and developed sites. Any pest management plan or activity must be formulated within the framework of the management zones in which it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

Weeds are usually described as any plants growing where they are not wanted. Any undesirable grass or broad leafed plant species, from a small herbaceous plant to a woody shrub, vine, or tree, may be considered a weed if it is growing in a landscape bed or in a structure.

Dicotyledonous (broad-leaved) plants are those that have two cotyledons in each seed. They are characterized by broad leaves and often have woody stems. Some species (e.g., sunflower) only become woody in old parts of stems and roots; these are referred to as semi-herbaceous dicots. Most weeds have little or no woody tissue (e.g., plantain, dandelion) and are herbaceous.

Grasses are members of the plant family Gramineae. All grasses are monocotyledonous and have long, narrow leaves with parallel veins and fibrous root systems. Some grasses produce underground stems called rhizomes (e.g., Kentucky bluegrass, quackgrass) or aboveground runners called stolons (e.g., creeping bentgrass), while others produce both (e.g., bermudagrass).

It is important to understand the distinction between monocots and dicots since the selectivity of many herbicides is based on which type of plant they kill. Thus many herbicides which would kill a dicotyledonous weed in turfgrass, which is a monocot, could not be used in a landscaped area since woody landscape plants are also dicots.

The description of each potential weed species is beyond the scope of this report. Contact the Cooperative Extension Service of the agricultural university in your state or your regional Integrated Pest Management coordinator for specific information on the most important weeds in your region. A list of useful pictorial weed guides is included in the reference section.

IDENTIFICATION AND BIOLOGY OF WEEDS OF DEVELOPED AND HISTORIC SITES

Undesirable plants (weeds) invade man-made environments such as landscape beds wherever they are established. Weeds are often found where soil has been exposed or disturbed by compaction, planting activities, or maintenance activities. They also occur where the turfgrass or groundcover is weakened by adverse environmental conditions, diseases, or pests to the extent that it cannot compete for nutrients, water, or light with weed species. Weeds are very common where the grass or groundcover species being grown is not well-adapted to its environment.

Life cycles

The life cycles of weeds can be grouped into the following major types.

Summer annual weeds. These weeds grow each spring or summer from seed. Examples include prostrate spurge, ragweed, large crabgrass, and goosegrass. They mature, produce seeds, and die in one growing season. Seeds generally overwinter before germinating the following spring. The majority of annual weeds are of this type. Some annuals, such as crabgrass, can root from leaf-stem junctions, forming dense colonies.

Winter annual weeds. These weeds (e.g., henbit, shepherdspurse, annual bluegrass) germinate in the fall or late winter from seed, mature and produce seed during the following spring, and die in early summer. Seeds of most of these species are dormant during the spring.

Indeterminate annual weeds. These weeds, such as chickweed and annual bluegrass, can germinate and grow during most seasons in certain regions.

Biennial weeds. These weeds may germinate at any time during the growing season. Examples include wild carrot, bull thistle, and mullein. They usually produce a radial cluster (rosette) of leaves lying close to the soil during the first season. In the second year they produce flower stalks (using food stored from the first season's growth), produce seeds, and die.

Perennial weeds. These plants live for three or more years. Some species may not flower the first year, while others may produce mostly nonviable seeds. Many perennials (e.g., curly dock, dandelion, and common milkweed) spread primarily by producing seeds, while others (e.g., field bindweed, white clover, silverleaf nightshade, bentgrass, and quackgrass) spread both by seed and vegetatively. The latter can occur by rhizomes, stolons, tubers, or rooting of stem nodes that touch the soil.

The seasonal abundance of weeds is related to their specific life cycles. Summer annuals grow from spring until fall, then are killed by low fall or winter temperatures. Winter annuals grow from fall to late spring, so they are usually not found during the summer. Biennials grow during the spring, summer, and fall of their first year, survive over the following winter, and flower during the next growing season. Therefore, some biennial stages are likely to be present at any time of the year. Perennials grow during each growing season. Their aboveground structures may die over the winter (e.g., yellow nutsedge) or may remain viable but dormant.

Knowledge of the life cycle of a particular weed species is an important part of its management. For example, mowing a patch of annual weeds to remove the flowers can prevent seed set. Refrain from cultivating areas where there are high populations of weeds that reproduce by rhizomes; this cuts the rhizomes into pieces and each piece can generate a new weed plant.

Impact of Weeds

The most obvious impact of weeds on turf areas is the competition and replacement of desired plants by weed species. In the case of weeds that overgrow an area and then die, such as crabgrass and knotweed, unsightly dead areas can be created. This often leads to the necessity for increased expenditures for turf maintenance. In landscape beds, weeds can grow among desirable plantings or among groundcovers and create an unsightly nuisance. This can lead to the need for hand weeding, which entails a high labor cost.

Toxicity to humans and animals is also a consideration. Some common weeds are poisonous if consumed (e.g., black nightshade, pokeweed, poison hemlock, and Johnsongrass); cause inflammation when touched (e.g., stinging nettle, poison ivy, oak, and sumac); or cause allergic reactions (e.g., common ragweed, goldenrod). Visitor injury or annoyance can result from bees or wasps seeking nectar from some weeds. Furthermore, many weed plants or their seeds have spines, thorns, or burs which can have similar allergic effects.

Weedy areas provide habitat for beneficial insects but may also attract rodents and arthropods such as rats, ticks, and fleas that might attack humans and domestic animals or carry diseases which will affect humans and domestic animals. Weeds can also serve as hosts for some fungal pathogens and insects which might attack desirable plants.

Weeds can also grow large enough to cover signs, block trails, or obstruct historic landscapes or vistas, interfering with visitor use of the park. Weeds that grow on buildings can cause structural damage if they grow into cracks in mortar or bricks; sometimes they will stain buildings as well.

Weed Habitats

Two habitats will be considered in this report; landscaped areas (where natural vegetation has been replaced or augmented with other plants, usually for aesthetic purposes) and buildings. Weeds growing in landscaped areas are found where soil has been exposed or disturbed by traffic or weakened by adverse environmental conditions, diseases, or pests to the extent that they cannot compete for nutrients, water, or light with weed species; where the desired plantings are not as well-adapted to their environment as are native or exotic weed species; and where the growth of the desired plants modifies their local environment so that natural ecologic succession to weed species occurs (in the absence of control).

Buildings, ruins, and other artificial sites can be considered disturbed environments, which will become populated by pioneer plant species if there is no intervention. Weeds can become established anywhere that a suitable substrate and water are found. Gutters, cracks in roofs, walls or foundations, and chinks in masonry all can provide suitable locations for germination of weed seeds. Woody plants can take root in soil pockets or deep cracks and crevices.

MONITORING AND THRESHOLDS FOR WEEDS AT DEVELOPED AND HISTORIC SITES

Regular monitoring for weeds is an essential part of a weed integrated pest management program. Weeds are most easily removed when they are small or present in low numbers; in the case of weeds which grow into structures or walkways it is important to remove them before serious structural damage occurs. In addition to monitoring for population density, identification of the species is important. The biology of the weed will often determine when it is to be removed or the most appropriate herbicide if chemical control is necessary.

Conduct weekly inspections around buildings and in landscape beds, recording weed species observed. Some estimate of density, such as number per square foot or number along a transect, should be recorded as well. If structural damage is already occurring, this should be noted as well. This type of information will help to correctly time weed removal. It will also help to prioritize areas for weed management if resources are limited and to evaluate the success of weed removal strategies used.

Certain areas are more likely than others to have high weed populations, and these should be the emphasis of your monitoring program. For example, recent cultivation will expose previously buried weed seeds to light. Heavy pedestrian traffic can lead to bare, compacted soil, which may be likely to support weed growth.

NON-CHEMICAL WEED CONTROL IN DEVELOPED AND HISTORIC SITES

The types of intervention strategies employed for management of weeds in the areas mentioned above will depend on where the weeds are located (landscape or structural), the size of the area in which the weeds are to be managed, the biology of the weed species present, the resources available for weed management, and the weed density that can be tolerated. Minimizing the spread of existing weeds and preventing the growth of new weeds should be the focus of a weed management program. One weed plant can produce hundreds of seeds which could potentially disperse over a wide area.

The objective of the site must be considered before selecting a weed control strategy and technique, especially in the case of historic sites. Filling and sealing the chinks in a stone wall might prevent weed growth, but that option is not available if it is not historically correct. Careless use of electric weed trimmers could damage fragile historic structures. Certain herbicides are corrosive and should not be sprayed near susceptible surfaces. The cultural resources staff should be consulted before implementing weed control in historic sites.

Weeds in the Landscape

Weeds in the landscape are generally considered to be unsightly and thus have a very low aesthetic threshold. In the case of new plantings, take time before establishment to remove existing weeds. In the case of existing plantings, emphasize the use of monitoring to detect weeds while they are still small or present at low population densities.

Weeds Around Buildings and Structures

Early detection and removal of weeds around buildings and structures such as benches and fences is especially critical. Once weeds grow into foundation cracks they become unsightly, difficult to manage and can do serious structural damage. This leads to high maintenance and repair costs. Weed growth into structures and on patios and walks can be partly prevented by proper maintenance of these structures. Filling of cracks in mortar and sidewalks so that organic debris cannot accumulate inside them will help to eliminate the entry and subsequent germination of weed seeds.

Weed Biology

Weed management must be based on a knowledge of the biology of the weed species. This is in turn dependant on correct identification of the weeds at a site. For instance, there is no point in applying a pre-emergent herbicide for crabgrass control if there is no crabgrass. It would also be fruitless to apply a pre-emergent herbicide that acts by preventing weed seed germination for control of established perennial weeds. Likewise, it could do more harm than good to cultivate a landscape bed for yellow nutsedge control in July after nutlets have formed; the cultivation will break the nutlets into small pieces and produce more weed plants. However, cultivation early in the season could remove the young plants before nutlet formation and might be an effective form of nutsedge management.

Physical Methods of Weed Management

Barriers and mulches are often used to eliminate a substrate in which weed seeds can germinate. While this is often a good, long-term solution to a weed problem, it is usually expensive to install. The elimination of the need for weed management may pay for the installation of the barrier over the long term, however.

One type of barrier would be the installation of paved walkways rather than soil, or the use of pavement or bricks under benches and around fences. This may not represent a permanent solution if cracks (and subsequent weed growth) are allowed to develop in the pavement. Depending on the site, it may be objectionable for aesthetic reasons as well.

Bare drainage ditches or pond banks can be lined with stones or desirable vegetation to help eliminate bare soil areas which are favorable for weed growth. This may not work in high-use areas where children could play with the stones, but might be a good solution to a weed problem in low-use areas of a park.

Weed mats are frequently used in landscape beds as a barrier to weed seedlings. These are made of materials which permit passage of air and water to plant roots but serve as a physical barrier so that weed seedlings cannot develop. While they are often effective, initially they are expensive to purchase and install. Also, weeds which grow through them cannot be pulled because the barrier will tear. For a complete discussion of the pros and cons of these materials, as well as a list of suppliers, see Billeaud and Zajicek (1989) and Lytton (1990).

Use of mulch in a landscaped areas is another common practice to reduce weed populations. This will not eliminate a problem, since weeds can grow through a mulch or germinate in it as it starts to decompose. A wide variety of material is available for use as mulch; the most appropriate mulch for a given situation depends on expense, effectiveness, aesthetics, availability, and types of plants growing in the mulched area. For example, plastic sheeting can be an effective mulch but it is unsightly and may pose disposal problems. Some stones or cinders may drastically alter soil Ph, while decomposition of sawdust or non-composted bark mulches can rapidly deplete soil nitrogen. For more information on the advantages and disadvantages of different mulch materials, as well as information on specialty materials which may be locally available, contact the Cooperative Extension Service at your land grant university.

Another type of mulch to consider is a living mulch. This involves the use of a groundcover to cover the soil around larger landscape plants. Sometimes this is supplemented with the use of a fast-growing annual to fill in bare areas between groundcover plants before they become large enough to cover the soil. Care must be taken not to use an invasive groundcover which may itself become a weed.

Mechanical Weed Management

Cultivation and hand-removal of weeds will be most cost-effective in small areas, eliminating small, newly established weed plants during seasons (usually the spring and fall) when the soil is moist and weeds are most easily removed. Keep in mind that there are certain times when cultivation will do more harm than good. Cultivation of annual weeds when mature seeds are on the plants is probably not a good idea, nor is hoeing of perennial weeds that regenerate by rhizomes or tubers after these structures have formed. Regular mowing is often sufficient to control weeds over large areas. In small areas, electric weed trimmers or propane burners are often used for weed control.

Biological Control of Weeds

Biological control of weeds in rangelands and waterways has been extensively investigated and seems to have a great deal of potential. This is not so for weeds in landscape settings, however. The only weed that would be found in a landscape that is currently under investigation as a biological control candidate is Canada thistle, *Cirsium arvense.* It is doubtful whether weed densities required for a biological control agent to be effective would be tolerated in a landscape. For more information on biological control of weeds, see Grossman (1989a) and Grossman (1989b).

CHEMICAL CONTROL OF WEEDS IN DEVELOPED AND HISTORIC SITES

When selecting a herbicide for use against a weed it is essential to identify the weed species, since many herbicides are specific in the types of weeds they kill (e.g., only grasses prior to germination, only broadleaf plants, most effective against poison ivy). Some herbicides are non-selective and will kill all vegetation whose leaves they contact; others are selective but are absorbed by roots of non-target plants and may injure or kill them as well. Mulgrew (1990) is a good resource for information concerning the use of herbicides in landscape beds. You should also contact your regional Integrated Pest Management coordinator or state Cooperative Extension Service for herbicide recommendations for your area, as well as for information on new herbicide formulations, since these change frequently.

Also consider that use of a non-selective herbicide for weed control may lead to an increase in weed problems in the future. The bare ground created in this situation could serve as a site for invasion by new weed species.

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Yellowjackets

This module is intended to serve as a source of basic information needed to implement an integrated pest management program for yellowjackets. Any pest management plan or activity must be formulated within the framework of the management zones where it will be implemented. Full consideration must be given to threatened and endangered species, natural and cultural resources, human health and safety, and the legal mandates of the individual parks. Recommendations in this module must be evaluated and applied in relation to these broader considerations.

The name "yellowjacket" refers to the typical yellow and black bands of color on the abdomen of a variety of wasps common to North America and elsewhere. Some species are actually black and white, but are known by the same name. Yellowjacket management is important because the vast majority of visitors regard yellowjackets (and other stinging insects) as a threat to their health and safety, as well as impediments to their enjoyment of the location.

In terms of management, it is important to remember that all varieties of yellowjackets are beneficial to the natural environment due to their predaceous behavior and consumption of large quantities of insect pests. If a nest does not pose a real, direct threat to personnel or to the public, it is recommended that you leave it alone.

The following information is intended to provide you with a description of various species of yellowjackets, their life cycle, habitat, and nest biology, as well as management issues in monitoring and control and medical treatment of stings. For more detailed information, you should consult the recommended reading list or discuss specifics with an appropriate professional. The medical advice offered is not intended to replace professional medical care. **Be prepared to handle emergencies resulting from stinging insects by contacting specialists in the area who can offer advise in an emergency situation. Locate these resources before the emergency occurs.** Someone's life may depend upon the level of preparedness.

Like any other creature, yellowjackets can be understood, managed, even enjoyed if you know a little bit about their behavior. Proper planning can avert most of the problems commonly encountered, thus increasing everyone's enjoyment of the outdoors.

YELLOWJACKET IDENTIFICATION AND BIOLOGY

Yellowjackets (*Vespula*, *Dolichovespula*, *Vespa*) are social insects that build enclosed paper nests underground, in trees, or in other structures above ground. Of the 19 species of yellowjackets and hornets found in North America, only five are considered pests. They are commonly referred to as eastern, western, southern, german, and common yellowjackets. These types are considered pests because they are all scavengers who come into frequent contact with humans as they forage for food. Remember, though, that all yellowjackets, including those considered to be pests, are actually beneficial insects in the natural environment due to their consumption of large quantities of other insects, many of which are also agricultural pests.

It is not necessary to identify the species of yellowjacket for purposes of management; therefore, such information is not included here. If more information is desired, refer to the bibliography. Akre et al. 1981 provides detailed taxonomic keys to the yellowjackets of North America.

Newly-produced queens are the only members of yellowjacket colonies to survive the winter (except in some situations in Florida.) From late March to May, they emerge from hibernation. Having been fertilized by males the previous autumn, the queen lays approximately 45 to 70 eggs, which hatch and become the first generation of workers. The queen continues to lay eggs, forage, and care for her brood. When the first five to seven emerge, they function as workers and care for all subsequent offspring. The queen does not leave the nest again. Workers feed the young, expand the underground nest by digging, produce paper comb, and protect the nest. Yellowjackets perform all duties of the nest at all ages. Yellowjacket workers are not sterile, but are kept from laying and caring for their own progeny by inhibitory chemicals (pheromones) produced by the queen. If the queen is lost, workers will produce male offspring.

Colonies grow slowly until mid-summer, when successive worker broods emerge and growth becomes exponential. Pest species typically have 500-5,000 workers at peak population. Increased foraging activity in areas frequented by human beings, coupled with a competition-induced aggressiveness and willingness to sting, leads to a sharp upsurge in the number of stings in late summer and fall (Davis 1978). By autumn, colony size has begun to decline. New queens mate and go into hibernation, while males leave to mate and die outside the colony. With the advent of cold weather, the old queen and workers die as well. Each year's population of yellowjackets in any given area will be affected by the weather, and thus will differ from year to year. Sudden cold snaps in the spring can sharply reduce populations for the rest of the year.

MONITORING AND THRESHOLDS FOR YELLOWJACKETS

Although scientists have developed very precise ways of monitoring insect populations, a practical approach tailored to the needs of your facility seems most advisable. If visitors and employees are not frequently bothered or stung by yellowjackets, it is reasonable to conclude that your management system is adequate and little, if any, monitoring need be done. However, if yellowjackets are becoming a nuisance, you may wish to take remedial action (as described herein) and develop a monitoring routine.

For example, you could select a specific garbage can (or other site that appears to draw a large number of the pests) and count the number of foragers that visit within a certain period of time (10 minutes). It would be best to monitor at roughly the same time each day. Take remedial action and see if improvements lead to a reduction in numbers. If stings have become a problem, you may also wish to keep track of the numbers of persons stung and correlate these figures with those resulting from forager monitoring. That is, at the point where stings are very infrequent (or approaching zero), that number of foragers in a given time period may be an acceptable number, implying good management of the facility. On the other hand, frequent complaints from visitors (or the observation by park personnel of pestered patrons) should be construed as requiring additional effort. Tolerance is expected to vary from one location to another. Remember, however, it is the great outdoors--some presence of yellowjackets is natural.

Contingency Planning for Yellowjacket Problems

Plan for emergency care of sting victims who are dangerously sensitive to venom. Have first-aid facilities or advice for non-sensitive victims. Monitor individuals who have received stings until you know they are safe. Pain or swelling at the site of a sting is a normal, non-threatening reaction; impairment of breathing, swelling of lymph nodes, dizziness, fainting, or similar extreme reactions are not normal and require expert help. Non-allergic individuals may find applications of ice, meat tenderizer or over-the-counter sting swabs helpful. If your facility wishes to assist sting victims with such remedies, include them in your first-aid kit. Be aware that only about 0.4% to 0.8% of the human population is seriously sensitive (in a life- threatening way) to wasp/yellowjacket venom. Many of these individuals already know of their allergy and carry sting kits or wear medical identification bracelets to safeguard themselves. Although anaphylactic shock can occur in as short as 10 minutes (and can cause coma or death), delayed reactions may occur up to 20 hours after the sting is received. (See Akre et al. 1981; Frazier 1976 for details). Also be aware that many individuals refer to themselves as "allergic" to stings, but do not present life-threatening symptoms--rather, they are referring to the normal swelling reaction one may get after being stung. Be sure that employees know how to question sting victims to accurately ascertain their reaction history and thereby determine whether emergency assistance is needed. An information sheet that may be helpful as a hand-out to visitors is included in this module.

Provide good public educational information on yellowjackets, other wasps, and bees at your location. To be most effective, these should include color drawings or photos that can be understood by visitors who cannot read English. Consider the advantage of making signs in both English and Spanish, or other languages common among your visitors. If possible, have knowledgeable staff available to answer questions that may arise from the information you provide.

If absolutely necessary, destroy structural nests, ground nests, and aerial nests with approved chemicals. This will be somewhat easier to do in dark or semi-dark conditions than in daylight. Be sure to dispose of all chemical containers in an environmentally-sound manner.

NON-CHEMICAL CONTROL OF YELLOWJACKETS

Non-chemical control of yellowjackets is achieved by reducing contact between humans and yellowjackets in every way possible. Displays, handouts, and other forms of communicating information should teach the visitor that stings are mainly avoidable by following certain precautions. Educational materials should emphasize the positive role wasps play in a healthy environment.

Sanitation

All refuse containers should be solid ones (no wire mesh, etc.) made of plastic or metal and equipped with wasp-tight lids to prevent foragers from gaining access to the interiors. All containers should be periodically checked for holes, cracks, etc., and repaired immediately. Refuse should be collected on a regular basis **before** containers are completely full. This may entail collection several times a day, particularly in picnic areas and during periods of heavy use of facilities, such as on weekends or holidays. Containers should be washed out regularly to reduce odors that attract yellowjackets. Use steam or soap if necessary, and hose down surrounding concrete areas as well. Plastic bag liners aid in sanitation and control of fluids that attract insects as well. Place trash cans as far as possible from picnic tables to reduce interactions between visitors and wasps. Monitor garbage cans for foraging wasps to determine local populations and spot critical areas for improvement.

Provide lids and straws on all soft drink containers sold by concessions. Be aware that yellowjackets can easily enter the openings of aluminum beverage cans if not carefully monitored by the user--and can thus present a hazard if accidentally ingested during the process of drinking from the can. For this reason, paper cups with lids and straws are safer.

Trapping

Trapping can at best provide only temporary relief in very limited areas, due to the large nest sizes of many colonies. It should therefore be considered secondary to the previously-mentioned management strategies.

Funnel traps using synthetic lures such as heptyl butyrate have been used successfully to capture western yellowjackets to a tolerable degree (Davis et. al 1973), but lures have not proven successful with the eastern species. A problem with synthetic lures is their inability to target only yellowjackets that are presenting a pest problem, while ignoring those performing their natural beneficial function.

Traps using raw fish as bait have been used to temporarily control *Vespula pensylvanica* (Akre et. al 1982). Cut the skin to expose the fish's flesh and suspend it above pans containing water and a wetting agent (like dishwashing soap) to reduce surface tension. Yellowjackets visiting these traps typically cut large pieces of flesh from the carcass and attempt to carry them to sites where they can chew them into smaller pieces. In so doing, they fall into the water and drown. Advantages of this method include ease of construction, effectiveness, and avoidance of toxic materials. Disadvantages include the need to change the bait frequently, as yellowjackets will not scavenge spoiled flesh, and the attractiveness of the bait to dogs, cats, flies, and other wildlife. Chicken-wire cages can be placed around traps to prevent disturbance by large animals.

In a 1974 test (Akre et. al 1982), nine traps set in a resort area captured nearly 1000 foraging workers per week. Trapping combined with improved garbage management reduced active foragers in the area to tolerable levels within two weeks.

Biological Control

Biological controls against yellowjackets are not currently known. Naturally- occurring parasites and predators exist, but have little or no effect on colony dynamics. Scientists continue to experiment in this area, giving hope of successful methods for the future.

Mechanical Control

Vacuuming of nests is the chief method employed, using a canister-type vacuum cleaner. One must be prepared to work quickly and be dressed with protective clothing (such as a beekeeping outfit). Avoid cutting into the nest, as this provides more than one exit for the angry wasps. Be prepared to immediately plug the entrance to the canister bag when removing it from the vacuum, as the yellowjackets inside will not be dead. The bag can then be frozen to kill the contents. If nests are removed from structures, the outside entrance should be sealed up, if possible, to prevent re-entry.

CHEMICAL CONTROL OF YELLOWJACKETS

Due to the large numbers of colonies and workers usually present in any area, wide chemical control of foragers is impractical, if not impossible. However, individual colonies located in hazardous spots can be selected for destruction by chemical means, if other methods are deemed unfeasible. Spraying should be attempted only after dark, when all foragers are back in the nest; in addition, the exterminator should wear a protective suit and take precautions against inhaling chemical fumes.

New products are continually introduced to control yellowjackets. It is recommended that you consult with an agricultural supplies dealer or your regional Integrated Pest Management coordinator regarding products appropriate for your needs. Do not use more than the recommended amount and use only for the purpose recommended on the product. Some products require the services of a certified pesticide applicator. Be sure to dispose of all empty pesticide containers in an approved toxic waste facility or container.

Do not use gasoline or other flammable liquids to destroy ground-dwelling yellowjackets. Doing so poisons the soil and can result in explosions or serious burns. Most chemical controls for yellowjackets are aerosol products containing pyrethrins, rotenone, and a cooling agent to lower nest activity and provide rapid knock-down.

Nests in structures are the most difficult to destroy. Do not simply plug the hole of a healthy colony, or the workers will chew a new hole through the wall and possibly emerge into human living space. For suggestions on how to treat such nests, see Akre et. al (1980) and Nixon (1982), and consult your regional Integrated Pest Management coordinator on the most effective way of exterminating a specific nest.

YELLOWJACKET FACT SHEET

Yellowjackets are small yellow-and black-banded wasps that build nests in the ground or paper-like nests in trees. The colony will reach maximum size in late summer. Worker yellow-jackets are common around picnic areas where they forage for food.

YELLOW JACKETS ARE ATTRACTED TO WAYS TO DECREASE STINGS

Perfumes and other scents Don't go barefoot Hairspray Don't swat with your hands Suntan lotion Use lids on soft drink cups Cosmetics Put tight-fitting lids on trash cans Sweet food Empty trash frequently

STINGS

In most people, a yellowjacket sting produces an immediate pain at the site of the sting. There will be localized reddening,swelling, and itching. Ice or analgesic creams often relieve the symptoms.

IF YOU ARE STUNG

1. Remove the stinger by scraping from the side (for bees)
2. Apply cold water or ice in a wet cloth
3. Lie down
4. Lower the stung arm or leg
5. Do not drink alcohol

Some people experience an **allergic reaction** to yellowjacket venom. Allergic (anaphylactic) shock can be fatal if untreated. Symptoms usually occur 10-20 minutes after a sting but may appear up to 20 hours later. If you experience any of the following symptoms after being stung, obtain medical aid immediately.

SYMPTOMS OF ALLERGIC REACTIONS	WHAT TO DO
Hives	Lie down; victim should not be moved
Widespread swelling of limb	Lower the stung arm or leg
Painful joints	Apply ice
Wheezing	Do not drink alcohol
Faintness sting and fingers under medical aid	Apply a wide cloth tourniquet between the heart (should be able to place 2 bands); release after 5 minutes Get

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