Arborist Study Guide
Plant Pest List

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BIRCH LEAFMINERS

Insect and Disease Laboratory • 168 State House Station • 50 Hospital Street • Augusta, Maine • 04333-0168

Birch is host to a number of leaf-mining sawflies. Two of the more common miners are *Messa nana* (Klug) and *Fenusa pusilla* (Lepeletier).

Symptoms and Damage

Some of the most prevalent, and widespread causes of browning of birch leaves in Maine has been by one or the other of these European insects. Damage is a result of feeding between the upper and lower leaf surface by the larval stages. Early symptoms of attack are tiny grayish or discolored blotches in the leaf tissue surrounding the eggs. Eggs of *M. nana* are laid along the serrated leaf margin and larvae mine toward the center of the leaf. Eggs of *F. pusilla* are laid away from the leaf margin and larvae mine toward the margin. Mines appear roughly ten days after the eggs are laid and are translucent green at first. As the irregularly shaped mines become progressively larger they turn brown and cause the leaves to become wrinkled and distorted.

While both species can be an aesthetic problem especially on ornamentals, only *M. nana* appears to impact forest stands where it is often associated with the birch casebearer, *Coleophora serratella.* The combination of these two species can result in tree stress and may hasten infestation by the more destructive bronze birch borer, *Agrilus anxius.*

Hosts

The predominant miner on white birch in recent years has been *Messa nana.* This species also attacks gray and yellow birch as well. *Fenusa pusilla,* on the other hand prefers gray birch or gray birch hybrids. The ornamental European white birch and its cut-leaf varieties are attacked by both of these sawflies. Trees growing in open, sunny areas such as around homes and along roadsides seem to be preferred and frequently suffer the heaviest browning.

Life Cycle

Whereas *M. nana* appears to have only a single generation in any one year there may be 3-4 generations of *F. pusilla.* Both sawflies appear to commence activity at roughly the same time each spring however. The small black sawflies, which somewhat resemble black flies, start crawling over leaves in late May, soon after the foliage develops, and lay eggs in the leaf tissue. Emerging larvae mine as described above. The small flat pale colored larvae have various dark spots as they mature and leave their blackish specks of frass (waste material) within the mines. Larvae eventually leave the mines, drop to the ground and pupate in the soil. Pupae of *M. nana* remain there until the next spring whereas those of *F. pusilla* produce a new generation of adults to start egg laying again within 10-14 days.

Control*

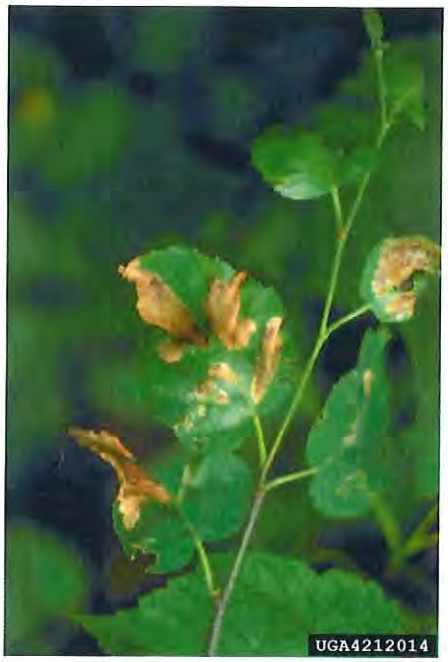
Good control depends on early, timely spraying for the first generation of sawflies and larvae to prevent appreciable numbers in any later generations. The best way to time the first application is to watch the new leaves as they first develop in May. Areas in leaves where eggs have been laid will develop a grayish cast rather than the normal green of the leaf. Developing mines will show up as tiny translucent spots of a lighter green color than the rest of the leaf when leaves are held up to the light. As soon as either the transparent spots or offcolor areas appear, foliar treatments should be applied thoroughly over the whole tree. A repeat spray may be necessary 10 to 14 days later if new symptoms appear. A number of insecticides including carbaryl, malathion, and acephate will all provide effective control against birch leaf miners, however, first check the insecticide label on the container for specific use and apply only in accordance to label instructions and precautions.

A systemic insecticide, disulfoton granular also provides excellent control against birch leaf miners. However, it is a restricted use pesticide to be sold only by licensed dealerships to certified pesticide applicators only. Disulfoton is applied as granules spread over the ground beneath the affected tree and thoroughly soaked with water in accordance with specific label use instructions and precautions. Disulfoton should be applied by mid-May to be most effective.

***NOTE**: These recommendations are not a substitute for pesticide labeling. Read the label before applying any pesticide. Pesticide recommendations are contingent on continued EPA and Maine Board of Pesticides Control registration and are subject to change.

Caution

For your own protection and that of the environment, apply the pesticide only in strict accordance with label directions and precautions.



Birch Leafminer Damage
Photo: Minnesota Department of Natural Resources Archives, Minnesota Department of Natural Resources, Bugwood.org

Birch Leafminer Adult
Photo: Whitney Cranshaw, Colorado State University, Bugwood.org

PURDUE UNIVERSITY

PURDUE EXTENSION

E-50-W

Landscape & Ornamentals

Department of Entomology

BRONZE BIRCH BORER

Timothy J. Gibb and Clifford S. Sadof, Extension Entomologists

Chlorotic leaves, sparse foliage and dying upper branches of white, paper and yellow birch trees are often the first visible symptoms of bronze birch borer damage. Closer examination will reveal ridges and bumps on the limbs and branches as well as occasional D-shaped holes in the bark. Peeling back bark from infested trees will further reveal irregular, winding, sawdust-packed tunnels. It is these tunnels that girdle the branches and eventually kill trees. Trees that receive no corrective measures usually die within a few years after the first dead branches are found.

LIFE CYCLE

The bronze birch borer is a serious native insect pest. The adult is a black beetle about 1/2 inch long and has bronzy iridescence on the back. It is a sun-loving insect and may be found crawling on the sunny side of the trunk during late May and early June.

The female deposits her eggs under cracks and crevices of the bark. Eggs hatch in 2 weeks or less, and the slender larvae tunnel immediately into the phloem tissue to construct their galleries. They may occasionally tunnel into the xylem (wood) to molt and overwinter. One or two years may be required to complete larval development. The larvae pupate in the xylem in late April or early May.

Bronze birch borer larva

Bronze birch borer adults

PREVENTING INFESTATIONS

By Maintaining Healthy Trees: Vigorously growing trees are less inclined to be damaged by borers than are trees in poor condition. Birch trees are relatively short-lived and thus old age, adverse weather conditions or other insect-related stresses can all contribute to weakened trees. Most ornamental trees are grown in lawns, which is not usually conducive to healthy, vigorous growth. In fact, lawn conditions are opposite the normal habitat of birch trees. Thus, birch trees are often in a weakened condition, making them attractive to the bronze birch borer.

By Using Resistant Varieties: One way to lessen chances of borer attack is to plant borer-resistant species like river birch, Japanese birch or the variety "Heritage". Another way is to maintain healthy trees by watering.

Birches in lawns should be watered every 7-10 days IF rainfall amounts are less than 1 to 1 1/2 inches per week. To water properly, allow the garden hose to run beneath the tree for several hours so that plenty of moisture reaches the root zone.

Prevent defoliation and injury by controlling other birch tree pests such as aphids, scales, and leafminers.

CONTROLLING EXISTING INFESTATIONS

Sanitation: Remove dying trees and branches before adults emerge in early May. This can reduce the number of borers available to attack your trees.

Insecticide Sprays: Between mid-May and mid-June, homeowners can spray infested trees with permethrin (Eight) or Spectracide Bug Stop to kill adults emerging from bark and feeding on leaves. Commercial applicators can spray Astro (permethrin) or Onyx (bifenthrin).

Systemic Insecticide Sprays: A soil drench of systemic insecticide between mid-May and mid-June can kill adult borers as they feed on leaves, or as they try to enter or leave the tree. Soil drench applications can kill young larvae that are already boring into trees. Homeowners may use dimethoate (Cygon, De-fend) or Imidacloprid (Bayer Advanced Garden Tree and Shrub). Commercial applicators may use commercial formulations of dimethoate or imidacloprid.

Bronze birch borer damage

Tunnels under bark (left) caused by bronze birch borer feeding will cause bark to appear bumpy (right)

Bronze Birch Borer: *Agrilus anxius* Gory

READ AND FOLLOW ALL LABEL INSTRUCTIONS. THIS INCLUDES DIRECTIONS FOR USE, PRECAUTIONARY STATEMENTS (HAZARDS TO HUMANS, DOMESTIC ANIMALS, AND ENDANGERED SPECIES), ENVIRONMENTAL HAZARDS, RATES OF APPLICATION, NUMBER OF APPLICATIONS, REENTRY INTERVALS, HARVEST RESTRICTIONS, STORAGE AND DISPOSAL, AND ANY SPECIFIC WARNINGS AND/OR PRECAUTIONS FOR SAFE HANDLING OF THE PESTICIDE.

Revised 5/2010

BROWNTAIL MOTH
Euproctis chrysorrhoea (L.)

History

Browntail moth caterpillar

The browntail moth was accidently introduced into Somerville, Massachusetts from Europe in 1897. By 1913, the insect had spread to all of the New England states and New Brunswick and Nova Scotia. The population then dropped, for reasons that are not entirely clear, until there was just a residual population limited to Cape Cod and a few islands off the Maine coast in Casco Bay. Occasional outbreaks occurred on the mainland during twentieth century until the 1990's when browntail became a perennial problem along the southern Maine coast.

Damage

The larval stage (caterpillar) of this insect feeds on the foliage of many hardwood trees and shrubs particularly: oak, shadbush, apple, cherry, beach plum, and rugosa rose. Larval feeding causes reduction of growth and occasional mortality of valued trees and shrubs.

While feeding damage may cause some concern, the primary human impact from the browntail moth is the result of contact with poisonous hairs found on the caterpillars. Contact of these hairs with human skin causes a rash similar to poison ivy that can be severe on some individuals. People can also experience respiratory distress from inhaling the microscopic hairs that blow around in the air.

Description and Life History

The browntail moth produces one generation a year. It has four life stages; egg, larval, pupal, and adult. The larval stage lasts for nine months, from August through June. In the fall, colonies of larvae build winter webs in trees constructed from a single leaf wrapped tightly with large amounts of white silk. A colony consists of 25 to 400 or more larvae. The larvae overwinter within two to four inch long winter webs situated far out on branch tips. Webs are found most often on red oak or apple trees.

Aside: Fall webworm nests, often confused with the browntail moth winter webs, are loose, further in on the branches and more often found in ash trees. Eastern tent caterpillar tents are found in crotches and forks of apple and cherry tree branches during the spring.

In the spring, as soon as the earliest leaf buds open, the larvae become active and crawl out of their webs to feed on the tender new leaves. They may devour the foliage as fast as it develops. For a time the larvae crawl back into the web at night, but as they become larger they remain out on the leaves. By late June, larvae are full grown. Large larvae, about 1 1/2 inches long, are dark brown, have a broken white stripe on each side of the body and conspicuous, two reddish spots on the posterior end of the back.

These should not be confused with larvae of the eastern tent caterpillar which has a single, solid, white stripe down its back or the gypsy moth which has paired blue and red spots on its back.

In late June, the larvae spin rough cocoons in which to pupate. Pupal cocoons are full of toxic hairs and should be removed from buildings or trees only with great caution. The pupae develop into moths which emerge from the cocoons in July. The moths have a wingspread of about 1 1/2 inches and are strongly attracted to light. Wings and midsection are pure white while the abdomen (rear part of the body) is brown with a conspicuous tuft of brown hairs at the tip.

After emerging, the females lay eggs in masses on the underside of leaves and cover the eggs with brown hairs from their bodies. Each female lays 200 to 400 eggs. The eggs hatch during August or early in September and the young larvae feed for a short time on the leaves before building their winter webs. This fall feeding does little damage to the trees.

Control

Non-chemical: Control of browntail moth populations in isolated areas may be obtained by clipping the overwintering webs and destroying these webs by either soaking in soapy water or burning them. This control should be undertaken in the winter and very early spring - September to mid-April.

Cocoons or caterpillars crawling on buildings can be removed with water from a high pressure hose.

Chemical: Webs and larvae are generally high up in trees and are difficult for a home owner to effectively control. Seek professional help from an arborist who is a licensed pesticide applicator if considering pesticide control. Pesticides should be applied when caterpillars are small and feeding; usually before the end of May. **Timing of pesticide treatment is critical, treatment before the end of May will prevent the development of the toxic hairs, treatment after the end of May will result in dead caterpillars and toxic hairs.** Undertake control measures as early as possible to reduce the exposure to the irritating caterpillar hairs. Contact a licensed pesticide applicator well before May to plan a control strategy. **Note:** A licensed pesticide applicator is required for applications of non-biological insectides to control browntail moth within the 250 of the high tide mark.

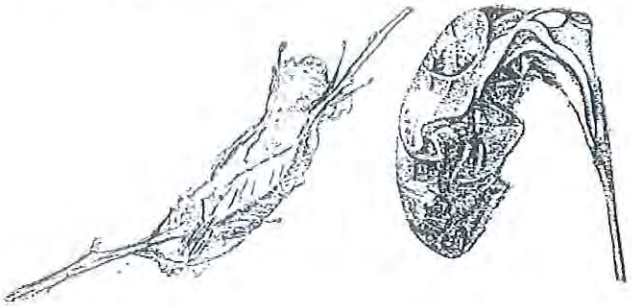
The Maine Forest Service maintains a list of pesticide applicators licensed and certified to treat browntail moths and other tree pests. It is available upon request by calling (207) 287-2431, emailing Patti.Roberts@maine.gov or on the website at:

http://www.maine.gov/dacf/mfs/forest_health/invasive_threats/browntail_moth_pesticide_applicator_info.htm

For more information on pesticides, both chemical and biological contact the Board of Pesticides Control at 207-287-2731

IDENTIFYING BROWNTAIL MOTH WINTER NESTS

The **browntail moth,** *Euproctis chrysorrhoea* (L.)*, overwinters as larvae (caterpillars) in colonies that are enclosed within webbed nests of white silk tightly woven around a leaf in trees or shrubs. The nests are spun in the early fall, contain 25 to 400 larvae, and remain firmly attached to twigs or small branches through the winter and early spring. The webs are often confused with silken structures formed by other less serious species of moths. Proper identification of browntail moth winter webs is important. Web counts can be used to quantify browntail moth caterpillar populations to be expected in the spring, a prerequisite for planning control projects. Also, *clipping and destroying overwintering webs by either soaking in water and detergent or burning in an incinerator can provide control of isolated populations located in low trees and shrubs.*



What browntail moth winter webs look like (image left):

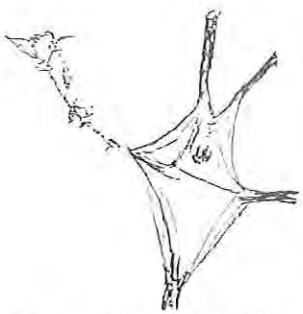
- 2-5 in. (5-10 cm) long
- white silk tightly woven around a leaf or leaves
- wrap of white silk tying leaf petiole to twig
- small brown hairy larvae within dense silk
- occur more commonly on twig ends of oak, apple, shadbush, cherry, beach plum, and rugosa rose

Among the webbed structures most often mistaken for browntail moth webs are old expired webs of the **fall webworm**, *Hyphantria cunea**. The webs, which may be numerous in some areas, are formed and occupied in summer and early fall by yellowish caterpillars with many tufts of long hairs and brown markings. The webs engulf the foliage on branch tips and can become 2 to 3 feet long. By mid fall the larvae have left the nests. During winter the nests become detached, and hang loosely from branches.

What expired fall webworm webs look like in winter (image left):

- variable length, 3-10 in. (7-20 cm)
- loosely tied matte of white silk and debris
- not associated with a leaf - any leaves caught in web do not have petioles tied to twig
- larvae not present in silk matte in late fall/winter/spring
- occurs on wide variety of deciduous hosts, especially ash and elm
- usually hanging from twigs or branches
- some webs fallen on ground in late winter

Remnants of expired webs or tents of the **eastern tent caterpillar**, *Malacosoma americanum** can also be mistaken for the winter webs of the browntail moth. These webs are built in the crotches of branches of wild cherry and apple trees in late April and May and may be a foot or more long when fully formed by early June. The tents also resemble the spring and summer webs of the browntail moth. Eastern tent caterpillars, which are present in their webs from late April to June, are covered with brown hairs have a solid whitish line down the middle of the back with a row of oval pale blue spots on each side. Browntail moth larvae are also covered with dark brown hairs, but have a broken white stripe down each side of the body and conspicuous, unpaired, reddish spots on the posterior end of the back.



Eastern Tent Caterpillar Spring Web

Eastern Tent Caterpillar Expired Web in Fall/Winter

*A detailed information and control sheet available on this pest from Maine Forest Service, Forest Health and Monitoring Division

Prepared by Maine Forest Service, Entomology Laboratory, 168 State House Station., Augusta, ME 04333-0168 Tel. (207) 287-2431

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Colorado State University
Extension

Cooley Spruce Galls

Fact Sheet No. **5.534** Insect Series | **Trees and Shrubs**

by W.S. Cranshaw*

Cooley spruce galls are commonly found on the new growth of spruce trees. These galls are produced by insects called Cooley spruce gall adelgids (woolly aphids). Galls appear early in the season as 2- to 4-inch cucumber-shaped growths. The galls are light green during late spring and early summer but dry out and become brown starting in mid-July. The galls often are mistaken for seed cones.

Cooley spruce galls are conspicuous and frequently cause considerable concern to homeowners. However, the galls usually do not cause any serious harm to the tree.

Extremely heavy infestations may cause minor retardation of tree growth and some distortion. However, in most cases, old galls are covered by new growth the following season and become almost unnoticeable a few years after they form.

Figure 1: Gall on blue spruce.

Life Cycle

Cooley spruce gall adelgids normally require two hosts (Douglas-fir and spruce) to complete their entire life cycle (Figure 4). Alternating between these plants, the life cycle often takes a year to complete. On spruce, woolly aphids overwinter as immature females underneath young branches. In the spring, females mature and lay several hundred eggs near developing buds. Eggs hatch at about the time of bud break and young nymphs migrate to the new spring growth. There they feed at the base of growing needles. Saliva introduced into the plant causes changes in plant development and produces galls.

The insects develop within chambers inside the galls and gradually increase in size. During the period when the insects are actively feeding, the galls stay green and are not readily observed. By midsummer, the galls dry out, the chambers open and winged forms of the insects emerge. These winged forms leave the original tree and most migrate to Douglas-fir trees. The abandoned galls continue to dry out and become increasingly noticeable a few weeks after the insects leave.

On Douglas-fir, eggs are laid on the needles and several generations of woolly aphids are produced. Yellow spots and bent needles result from feeding damage. The trees appear speckled with tiny cotton-like forms of the insect. No galls are produced on Douglas-fir. Late in the summer, some of the woolly aphids develop wings and fly back to spruce to deposit eggs, which produce the overwintering population. Others are wingless and remain on Douglas-fir trees, where they produce other overwintering forms.

Certain patterns of infestation are commonly observed on spruce. Most galling typically takes place on the more shaded sides of trees (north, east). Also, individual trees appear to vary greatly in their susceptibility to galling. Some trees, usually those that have a greener coloration, tend to be more heavily infested than other trees. This suggests

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Quick Facts

- Cooley spruce galls are common and conspicuous on blue spruce. However, they do little or no harm to the tree.
- The galls are produced by aphid-like insects, Cooley spruce gall adelgids.
- Cooley spruce gall adelgids require two hosts to complete their normal life cycle: spruce and Douglas-fir.
- Once the galls begin to form, insecticides usually are ineffective because the insects are protected within the galls.
- The best times to spray are in the spring before new growth starts or in the fall when over-wintering stages of the insect have returned to the tree.

www.ext.colostate.edu

Figure 2: Overwintered females.

Figure 3: Gall cross-section showing developing insects.

that resistance to galling is a widespread characteristic among spruce trees.

Control

Control of Cooley spruce gall is rarely needed to protect tree health. Infestations of this insect are highly cyclical, with their numbers often changing greatly from season to season. However, insects may be controlled to prevent aesthetic injuries that can detract from tree appearance.

Control must occur before galls begin to form. Apply treatments in fall, after the overwintering females have settled on the plants, or, most commonly, in spring. Spring applications are most effective if made before the insects have begun to swell with eggs, which typically occurs in late April.

Foliar treatments of carbaryl (Sevin) and permethrin have been most effective in Colorado State University trials. Horticultural oils have also been very effective but can cause temporary discoloration of spruce needles. See fact sheet 5.569, *Insect Control: Horticultural Oils*. Insecticidal soaps are only moderately effective on spruce, but are used widely to control this insect on Douglas-fir. Direct all foliar applications at the underside of spruce terminals where the overwintering aphids are concentrated.

Soil injections of imidacloprid can control Cooley spruce gall adelgids. However, the treatments often fail to kill many of the insects until after the galls are formed. These galls usually remain green rather than browning in early summer. Fall applications are recommended for this insect.

Removal of old galls will not affect infestations because the insects have left the tree by the time galls turn brown and become conspicuous. Old galls are not later used by any stages of this insect.

When establishing plantings, avoid placing Douglas-fir and spruce close together.

Table 1. Insecticides for control of Cooley spruce gall adelgids.

carbaryl (Sevin)
horticultural oils
imidacloprid (Merit, etc.)
insecticidal soaps
permethrin (Bug Stop, Astro, etc.)

Always read and follow mixing and usage instructions on the label. Direct applications against the overwintering insect, preferably before egg production has begun. Treatments usually are best made during warm periods in fall (mid-October or November) or in late March through April.

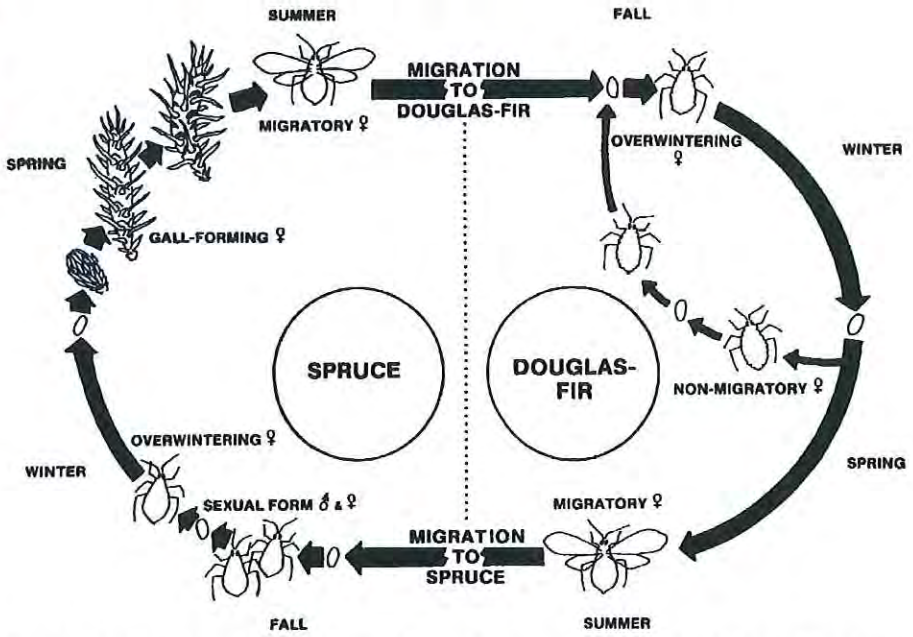

Figure 4: Life cycle of the Cooley spruce gall adelgid. Research indicates that the adelgids leaving spruce must develop on Douglas-fir before returning to spruce. Observations in Colorado suggest that spruce to spruce movement also may occur.

United States Department of Agriculture Forest Service Northeastern Area NA-FB-37-February 1990

The Eastern Tent Caterpillar

The eastern tent caterpillar is often mistaken for the gypsy moth. Though they are similar in appearance, they differ in habits.

The fully grown eastern tent caterpillar is about 2 inches long, black with a white stripe along the middle of the back and a row of pale blue oval spots on each side. It is sparsely covered with fine light brown hairs.

The gypsy moth caterpillar, when fully grown, is also about 2 inches long, but it has pairs of blue and red spots on its back. Compare the photos in Figures 1 and 2 to see the difference.

Figure 1. Eastern Tent Caterpillar.

Figure 2. Gypsy Moth Caterpillar.

Unlike the gypsy moth, the eastern tent caterpillar can be readily identified by the tent it constructs in the forks of tree branches (see Figure 3).

Tent caterpillars spend the winter in egg masses that are in shiny brown bands around twigs (see Figure 4).

Figure 3. Figure 4. Figure 5.

The gregarious caterpillars hatch in the earlly spring about the time tree buds start to open, and soon they begin to spin their silken tents in the branch forks (see Figure 5). The tent protects them from predators, such as birds, and from temperature extremes. Enlarging the tent as they grown, the caterpillars leave only to feed, usually at night.

The eastern tent caterpillar is found most often on apple and wild or ornamental cherry, and occassionally on pecan, hawthorne, beech and willow. When abundant, caterpillars will eat all the leaves, weakening, though seldom killing a tree.

Leaf-feeding can be prevented on small trees by destroying tents with a stick or pole, exposing the caterpillars to birds. Another preventive method is to prune the egg masses from twigs before the early spring hatch.

For more information, contact your county extension agent or the State Forester.

Authors:

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Daniel Twardus, USDA Forest Service

For additional information, contact:

USDA Forest Service
Forest Health Protection
180 Canfield Street
Morgantown, WV 26505
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USDA Forest Service
Forest Health Protection
271 Mast Road
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(603) 868-7704

USDA Forest Service
Forest Health Protection
1992 Folwell Avenue
St. Paul, MN 55108-1099
(612) 649-5261

Fact Sheets - Forest and Eastern Tent Caterpillars

Pest Management Fact Sheet #5022

Forest and Eastern Tent Caterpillars

(Malacosoma disstria) & (Malacosoma americanum)

James F. Dill, Pest Management Specialist
Clay A. Kirby, Insect Diagnostician

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Forest Tent Caterpillar

Description & Biology

The forest tent and eastern tent caterpillars are similar in habits and appearance. Fully grown forest tent caterpillars (larvae) have keyhole-like, or shoe print-shaped, whitish spots on each body segment. This fine line of spots runs down the middle of the back for the entire length of the body. On each side there is a blue line, which gives them a bluish appearance. Their favorite hosts are oak, poplar, maple and birch. The caterpillars do not build nests but leave mats of silken threads on trees where they travel or rest.

Nest

The eastern tent caterpillar differs in that it has a solid whitish line down the center of the back and an orangish stripe on the sides. They feed on apple, crabapple and cherry trees and build distinctive nests in forks of trees.

Eastern Tent Caterpillar and larva

The tent caterpillars lay their overwintering eggs (masses with a dark brown shellacked appearance) around small branches about the size of a pencil. Hatching occurs as buds swell and open, the young caterpillars feed on the buds, and the nests become apparent in late May. As the larvae grow they begin to feed on leaves. When the population increases, it is not uncommon for trees and forests to be defoliated. The caterpillars mature in the first part of June, with adult moths appearing during the last part of the month, when egg laying takes place. There is one generation per year.

Forest tent caterpillars have denuded forests in many areas for up to five years in a row. Trees can usually survive such defoliation, but with reduced growth. Disease, other insects, nutrition, moisture and general poor condition may cause tree mortality in defoliated trees. Higher defoliating populations of forest tent caterpillars typically drop after two years due to natural conditions, parasites, predators, and diseases. Freezing weather shortly after eggs hatch kills large numbers of caterpillars, and excessively high temperatures later in the spring kills large numbers of adults and reduces the viability of newly laid eggs.

anagement

Management for the eastern tent caterpillar and forest tent caterpillar is similar, except nests of eastern tent caterpillars can be removed and destroyed. Egg masses on branches of small trees around the home can be

removed and destroyed before hatching.

Organic methods include using B.t. (Bacillus thuringiensis), spinosad, insecticidal soap (be careful of leaf burning) or neem against small larvae. Other insecticides include carbaryl (Sevin) and malathion.

When Using Pesticides

ALWAYS FOLLOW LABEL DIRECTIONS!

Pest Management Office
491 College Avenue, Orono, ME 04473-1295
1-800-287-0279 (in Maine)

Back to Fact Sheets

UMassAmherst

The Center for Agriculture, Food and the Environment

Home of UMass Extension & Mass. Ag. Experiment Station

Published on *Center for Agriculture, Food and the Environment* (https://ag.umass.edu)

Elongate Hemlock Scale

Pest: *Fiorinia externa* (Ferris)
Order: Hemiptera
Family: Diaspididae

Host/Range

Elongate hemlock scale (EHS) is primarily found infesting eastern hemlock (*Tsuga canadensis*) and Carolina hemlock (*Tsuga caroliniana*), along with fir and spruce in Massachusetts. It may also be found in cedar, Douglas fir, pine, and yew when those plants are grown nearby an infested hemlock. Elongate hemlock scale is an armored scale, non-native to the United States and first observed in New York in 1908. This invasive insect from Asia (native to Japan and China) has since moved throughout the state of Massachusetts and parts of the coastal Northeast in landscaped as well as forested areas.

escription/Life Cycle

Adult female EHS are covered with an orange-brown waxy coating (test) that is approximately 1/20th of an inch long and longer than it is wide. The adult female and her eggs are yellowish-orange in color, as is the crawler or mobile stage. The immature males are slightly smaller and coated with a whitened-waxy test, which can sometimes be thread-like in appearance. Adult males are tiny, winged, and mobile and fly to the stationary adult females to mate.

Multiple life stages may be present throughout the growing season. Egg hatch generally occurs by the end of May or beginning of June, leading to the movement of crawlers from underneath the female's test (waxy covering), which settle at the underside of host plant needles, where they feed using their piercing-sucking mouth parts. Males and females mature, and the winged males then seek out matured females to mate. These mated females produce further eggs beneath their hardened coverings, and many life stages may overwinter. However, this insect primarily overwinters as eggs or fertilized females. These overlapping life stages can make management difficult. In the Northeast, one generation per year has been observed, whereas warmer areas in the southeastern United States may experience two generations per year.

Images


[1]


[2]

ımage

EHS damages the host by inserting its piercing-sucking mouthparts into the needle tissue and sucking out the fluids of the plant. This can cause chlorosis of the needles, premature needle loss, branch and limb dieback, and may also contribute to the eventual death of the tree, especially in the presence of other pests or when populations are high.

Management

Chemical management options can include foliar applications of horticultural oils, if complete coverage of the insect can be achieved (on smaller plants). These applications are usually more effective if made while crawler stages are present and exposed (not covered and protected by the waxy scale). Multiple applications per growing season may be required for management of this insect. A dormant oil application may be made between the end of March and early-April. A second application of horticultural oil according to label rates may then be made between late-May and early-June. Follow label instructions to reduce the risk of phytotoxicity when making any applications. Insecticidal soaps may be used, but since they dry quickly, it is difficult to time applications accurately so that they will impact the scale when it is in the vulnerable crawler stage.

On larger landscape specimens where complete coverage from foliar applications is difficult to achieve, systemic insecticides are available for management. Home owners will need to hire a licensed pesticide applicator to apply these materials. The systemic insecticide, dinotefuran, a neonicotinoid, can be effective within weeks of application. Application can be done as a trunk injection, soil drench, or basal bark spray. Bark sprays for elongate hemlock scale have been reviewed with success, although yearly applications may be necessary to achieve management of this insect. Dinotefuran can also protect trees from hemlock woolly adelgid for about 2 years. Although neonicotinoids are currently under review, particularly in reference to their potential impacts on pollinators, hemlock is primarily wind pollinated. For this reason, there is a reduced risk to bees and other pollinators, since there may be little to no pollen or nectar that would be attractive to beneficial insects (in some cases, pollen and nectar have been found to contain traces of systemic insecticides). Please note that another neonicotinoid, imidacloprid, does not manage elongate hemlock scale populations, but is effective against hemlock woolly adelgid.

As with all chemical insecticides, use best management practices to reduce drift, avoid applications during times of day when pollinators are most active or when flowers are in bloom, reduce impacts to non-target organisms, and observe all regulations when applying near schools, wetlands, and other sensitive areas or populations.

Management of elongate hemlock scale can be difficult to achieve, as all life stages of this armored scale insect can be present throughout the growing season, many of which are protected by the hardened scale covering. Multiple chemical applications are often necessary to keep populations down. Follow-up monitoring after chemical treatments should include using a hand lens or microscope to view the scales to determine whether live insects are present. Fertilizing trees with elongate hemlock scale, particularly with nitrogen-based fertilizers is discouraged. The added nitrogen may lead to population increases of the scale.

In some cases, highly infested landscape trees may be removed and disposed of away from healthy host plants. This option may be desirable for those who do not wish to make multiple chemical applications to manage this insect.

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UTAH PESTS fact sheet

UtahState University COOPERATIVE EXTENSION

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Fall Webworm

Ryan S. Davis, Arthropod Diagnostician, and Vincent P. Jones, Extension Specialist

DID YOU KNOW?

- Fall webworms are a common pest in urban and forest areas from mid July through August.
- Fall webworms can feed on over 100 species of trees, but cottonwood and chokecherry are preferred hosts.
- While unsightly, fall webworm damage rarely causes serious stress to trees and control is usually not recommended.
- Webs can be removed from low-growing trees by hand, with a pole or other apparatus, or pruned out. *Bacillus thuringiensis* is an effective, low-toxicity chemical option.

INTRODUCTION

The fall webworm, *Hyphantria cunea*, is a common defoliator of ornamental and fruit trees in Utah. Starting around late July, the caterpillars, webbing, and damage become noticeable, particularly in some of the canyons adjacent to populated areas (Little Cottonwood canyon, Logan canyon, Provo canyon, etc.).

Fig. 1. Adult fall webworm (*Hyphantria cunea*) laying eggs on the underside of a leaf.[1]

Fig. 2. Fall webworm (*Hyphantria cunea*) larvae feeding gregariously inside of a silken "tent."[1]

GENERAL BIOLOGY

Fall Webworm

Scientific Name: *Hyphantria cunea.*

Range: Native to North America.

Hosts: Cottonwood and chokecherry are preferred, but over 100 species can be consumed.

Identification Adult: The adult moth has a wing spread of 1 to 2 inches and is primarily white (body and wings), but can have black spots on the wings (Fig. 1). Occasionally, the legs and body will have orange markings.

Identification Immature: Full grown larvae are about 1 inch long, and have highly variable coloration (Fig. 3). Larvae are covered with long tufts of hair coming from orange and black bumps on the back (Fig. 3).

Life History: Adult moths emerge from June to July, mate, and lay eggs on the under sides of host leaves (Fig. 1). Eggs hatch in about 1 to 2 weeks; larvae mature in about 6 weeks and drop to the ground to pupate. Pupation sites frequently occur in the soil, leaf litter, or under loose bark. Moths overwinter as pupae. There are one to two generations per year depending on location.

Key Habits: Newly hatched larvae make a web "tent" around themselves and their food, which increases in size as they progress. Young larvae feed gregariously (in a group, socially) skeletonizing leaves (Fig. 4), and will consume whole leaves when older. Damage usually does not cause serious stress to the tree.

CONTROL

Outbreak populations of fall webworms seldom occur, and because they defoliate trees late in the growing season they rarely cause life-threatening damage to trees. Chemical control of fall webworms is usually not recommended. On occasion, high numbers of caterpillars may be unsightly and become a nuisance pest that warrants control.

Cultural and Physical Control Methods:

If webworms or their tents are located within a safe reaching distance by hand or pole, they can be physically removed or pruned out of the tree. A strong spray from a high pressure hose can also dislodge tents. Burning tents is not recommended, as this can cause much more damage than the webworms would inflict.

Chemical Control Methods:

Chemical control of fall webworms can be difficult because of their location in the tree (generally found high in the canopy) and because their webbing reduces the penetration of pesticides to the point that the insect may not come in contact with a lethal dose. When applying pesticides focus on the foliage surrounding the current tents or caterpillars to stop their expansion. When taller trees are treated, a high pressure spray gun should be used to obtain good coverage.

Fig. 3. Fall webworm (*Hyphantria cunea*) larvae can have great variability in their coloration.[1]

Fig. 4. Fall webworm (*Hyphantria cunea*) larvae feeding gregariously on a leaf--notice the skeletonizing pattern of the damage.[1]

The microbial insecticide B.t. provides good control of fall webworm with minimal impact on other insects or wildlife. Dipel and Bonide are two products that contain B.t. For best results apply B.t. when newly hatched caterpillars first appear.

In Utah there are over 140 products containing 29 different active ingredients (AI's) labeled for use on ornamental shade trees for fall webworm control. Examples of AI's include: acephate (1B), bifenthrin (3A), carbaryl (1A), chlorpyrifos (1B), imidacloprid (4A), premethrin (3A), and thiamethoxam (4A). The number and letter combinations following AI's indicate its chemical group. These groups are chemically similar insecticides that kill the target organism in a similar way, i.e., mode of action. Rotating insecticides with different chemical groups on a yearly basis can help reduce insect resistance build-up to an insecticide. For more information on insecticide mode of action and insecticide resistance, visit irac-online.org. Make sure the product you select contains the site, e.g., ornamental fruit or shade trees, to which you are going to apply.

PHOTO CREDITS

1. Lacy Hyche, Auburn University, Bugwood.org.

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GALLS OF MAPLE TREES IN OHIO

ENT-60
Last Updated: 04/16/2015

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Galls are plant structures that form as a result of arthropod feeding. The abnormal plant growth is stimulated by a combination of plant hormones and hormones introduced into the plant during arthropod feeding. The plant responds by increasing the production of leaf tissue, thereby enclosing the individual mite or insect. The enveloped arthropod feeds and reproduces from within the gall.

The upper surface of silver and red maple leaves often become covered with small, red, round wartlike structures about 1/16- to 1/8-inch in diameter. These are maple bladder galls caused by a small eriophyid mite, *Vasates quadripedes* (Shimer). The structures are generally noticed first in May, about the time the leaves have become fully expanded. At first the galls are green, but they quickly turn pink to red and eventually black. Leaves frequently become so covered with the galls that they completely twist out of shape and may even drop early.

On sugar maple, another leaf gall is commonly found, the maple spindle gall. This gall is caused by the mite, *Vasates aceriscrumena* (Riley). The gall appears as thin, elongate bladders arising from the upper leaf surface. These galls rarely distort the leaf, but considerable numbers of galls can make the leaves unsightly.

Maple bladder gall.

Maple spindle gall.

The maple velvet erineum gall is another gall commonly found on maples, primarly silver, Norway and boxelder maples. This colorful gall is caused by another eriophyid mite, *Eriopyhes* spp. Crimson to yellow, velvet-like patches form on the upper and lower leaf surfaces as a result of mite feeding activity. These conspicuous, hairlike patches may resemble a fungus or a disease.

The maple gouty vein gall midge attacks only sugar maple and causes thickened pouches along the major veins. These galls can completely crumple the early leaves of maples, often making them look like herbicide damage. The galls are caused by the larvae of a small gnatlike midge, *Dasineura communis* (Felt).

Maple velvet erineum gall.

Gouty vein gall.

Homeowners often become quite alarmed when they discover that their maple tree leaves are infested with leaf bladder, spindle, erineum or gouty vein galls. They fear that the tree is going to die unless something is done quickly. This is not the case. Maple leaf galls seldom, if ever, cause permanent injury to a tree, but they do detract from the beauty of the leaves.

Damage

Other than the mere aesthetic damage and possible early leaf drop, no significant damage is done to the health of maple trees. Following a mild winter, damage from these leaf galls can be excessive, but affected trees often send out new leaves to replace the damaged ones.

Description and Life Cycle

Both the maple bladder, maple spindle and erineum gall mites overwinter as free-living mites under loosened bark and around the callous growth of wounds, scars and pruned branches. These overwintered forms produce the gall-forming stage in early spring. When the maple leaves first appear, the mites migrate to expanding buds and begin to feed on the undersurface of leaf buds. This causes the formation of a blister, which expands into a hollow bladder or spindle as the leaf expands. The mites enter the cavity and continue to feed within its protective walls. This stage reproduces asexually within the galls, and the new mites mature by late June to mid-July. At this time, the galls dry out and the adult mites emerge. These mites then seek out overwintering sites.

The maple gouty vein gall midge spends the winter as full grown larvae in the ground and leaf litter under their host sugar maples. In late January into early March, these larvae spin small white cocoons in which to pupate. The pupae rest until April and early May when the gnatlike adults emerge. These midges have black wings and heads, but the body appears reddish from the eggs inside. Each female may lay up to 100 eggs among the leaf hairs on the lower leaf surfaces of expanding leaves. The tiny, maggotlike larvae hatch in a couple of days, and they migrate to the upper leaf surface. Here they line up in small groups along the major leaf veins. At these congregation points, the leaves swell and the vein edges fold over to form the galls. Within a few days the galls are fully formed. The larvae feed within the protection of the gall until October. At this time the galls dry and a slitlike opening is formed. The mature larvae drop to the soil to seek shelter.

Control Hints

Since these leaf galls of maple are typically only aesthetic and do not cause any real harm to the trees, control measures are generally not recommended. Tree owners and tree managers are encouraged to learn about the life cycles of these pests and learn that no lasting damage will result.

Strategy 1: Resistant Varieties

Norway maples and some of the named cultivars of maples with outstanding red or yellow leaf color appear to be resistant to these gall mites and midges. Talk to the plant supplier to see if the tree cultivar has a history of leaf gall problems.

Strategy 2: Cultural Control

Encourage plant health with proper fertility and adequate moisture. Prune out affected branches and twigs to reduce populations. Dispose of infested plant tissue in plastic bags to minimize spread of adult mites.

Strategy 3: Dormant Oils

Use of dormant oils on maples is discouraged because leaf and twig damage can result unless the tree is truly dormant. However, some reports of success have been made where the trunk has been drenched with dormant oil to kill the overwintering stages of the bladder gall and spindle gall mites.

Strategy 4: Standard Insecticide/Miticide Sprays

Several insecticides and miticides are registered for control of gall mites (eriophyid mites) and gall midges. If these products are to be used, they have to be applied precisely when the new leaf buds are opening. Most sprays have little, if any, effect because the window of opportunity is very short. Once the gall has formed, it is too late to make an application as the pest is protected by the gall and does not come into contact with the pesticide.

Strategy 5: Systemic Insecticides/Miticides

Several systemic pesticides (sprayed, soil-injected or trunk-injected) have been recommended as useful in controlling these gall-forming pests. However, little evidence of success has been found in the current literature. Once the gall has formed, it is too late to make an application. Always read and follow the pesticide label.

This fact sheet is a revision of HYG-2004.

A Horticulture Information article from the Wisconsin Master Gardener website, posted 25 May 2009

Maple Gall Mites

Bumpy growths caused by maple gall mites cover silver maple leaves.

If you have a maple tree, you may have noticed weird growths on the leaves in some years. These strange swellings are plant galls caused by the feeding of tiny eriophyid mites on newly developing foliage. The mites, less than 2mm long, are too small to be seen without magnification. The adult mites become active very early in spring, just as buds begin to open. As they feed on the developing leaf tissue, growth regulating chemicals produced by the mites interact with plant hormones causing an abnormal stimulation of cell growth. This abnormal growth forms the galls that gradually enclose the mites. Each mite species produces slightly different substances, resulting in the different characteristic gall shapes. The mites get nutrients from the inner gall tissue and are protected from natural enemies (and also pesticides).

Once the leaf has expanded completely and reached its full size, the adult mites lay numerous eggs within the galls and die. The eggs hatch into whitish-orange, carrot-shaped nymphs with two pairs of legs. They remain within the galls, feeding on the gall tissue until they have matured into adults, usually within a few weeks. They then exit through an opening on the underside of the leaf to crawl to newly developing leaves and begin the cycle again if the tree still has new leaf growth. Once the tree has stopped new growth, usually in early summer, the mites are unable to stimulate the abnormal growth to produce the galls. At this time they head for the trunk and branches to overwinter in roughened areas on the bark, bud scales, or other protected places. There may be several generations during a growing season.

Mite feeding causes the abnormal cell growth that causes gall formation.

Heavy infestations may cause leaf deformity.

The extent of infestations can vary greatly, with large concentrations of galls on some trees or portions of trees, and very little on adjacent trees or the other part of a single tree. The leaves closest to overwintering sites are most heavily infested. Abundance also varies greatly from year to year, with galls appearing sporadically. Heavy infestations may deform the leaves or cause premature yellowing and drop. But usually the mites and their galls cause little harm.

Homeowners are often concerned when they notice the highly conspicuous and sometimes colorful growths, and want to do something to correct the problem. Although the galls may look dramatic, they actually have very little effect on plant health, even in severe infestations. Control is generally not recommended for this primarily cosmetic problem on established trees. However, on newly planted trees with less foliage, galls may be a significant stress. Damaged leaves can be handpicked to reduce mite populations. Once the galls have formed, the mites are protected within the galls, so sprays will not be effective. For trees with extreme infestations, preventative treatments in very early spring to control overwintering mites – as they begin feeding on the expanding leaves but before the galls have enclosed them – may reduce the incidence of galls, but are generally of limited value. Application to the lower leaf surface should be made on a warm day in the spring when the leaves are about 1/4 expanded and again 10 days later. Because the mites overwinter on the host trees, dormant oil sprays can be quite effective (although oil sprays should not be used on sugar maple, Japanese maple and other oil-sensitive trees such as birch, hickory and walnut because of phytotoxicity problems). These should be applied prior to bud break. Because pesticides have limited impact on the mites, it is better to put efforts into good horticultural practices, such as watering and fertilizing, as needed, rather than spraying.

Infestations are often worse on leaves nearest the overwinter sites on the bark.

There are several different species of mites that cause different types of galls. The three main types are:

Maple Bladder Gall

These irregular, rounded swellings on the upper surface of the leaves begin green, then turn pink to red and finally change to black. The hollow, 1/16 to 1/8 inch structures each containing a single mite are held up on short "stems." They tend to be more numerous on early spring foliage, especially near the trunk and larger branches. When mite populations are high, the galls may cover nearly the entire upper leaf surface. Under these conditions the leaves are seriously deformed, tending to "cup" and fall prematurely. This type of gall is caused by the mite *Vasates quadripedes*, which attacks silver maple more commonly than red maple. Distribution of the mites on a tree tends to be irregular, and more solitary on red maple than on silver maple.

The round galls of maple bladder galls.

Spindle galls (on another plant, not maple).

Spindle Gall

These small, elongated, spindle-shaped growths generally occur on the upper leaf surface. The 1/5" long galls are about the same diameter as a pencil lead, tapering at both ends. They begin a green color and eventually change to tan. Caused by the mite *Vasates aceris-crumena*, this type of gall is found most commonly on sugar maple, but also on red, silver and whitebark maple.

Erineum Gall

Velvety, felt-like patches, formed of numerous microscopic galls amid the leaf hairs on the underside of the leaves, occur on sugar, silver and red maple, and occasionally Norway maple. The ¼-3/8" mat-like galls begin light green or yellow-green and often become more conspicuous when they change to a bright red or crimson. Several species of *Eriophyes* can produce this type of gall. A similar type of gall caused by *E. negundi* occurs on boxelder, and other mites cause this type of gall on different hardwoods.

Furry patches of erineum galls.

There is another less common, but striking, gall that may occur on red maple and sometimes sugar or silver maple. Unlike the three other galls, this one is caused by a midge (a type of small fly). Eyespot midge galls have bright red and yellow rings around the spherical growth.

– *Susan Mahr, University of Wisconsin - Madison*

Additional Information:

- Maple Gall Mites – UWEX publication A2691 at learningstore.uwex.edu/pdf/A2691.pdf
- Maple Bladder Gall, Spindle Gall and Gouty Vein Gall – Ohio State University Extension Fact Sheet HYG-2004-95 at ohioline.osu.edu/hyg-fact/2000/2004.html

Forest Health Fact Sheet
Gypsy Moth

Identification

The gypsy moth, *Lymantria dispar* (L.), gets its name from a behavior of its larger caterpillars, which generally migrate each day from the leaves and down the branches and trunk to rest in shaded spots on the tree or objects on the ground. Tiny, black young caterpillars are windblown to their food plants, where they will feed day and night but older stages of the caterpillars feed only at night to avoid drying out in the hot sun or being eaten by predators. Mature gypsy moth caterpillars are dark and have dark hairs, but you can clearly see the five pairs of dark blue spots and six pairs of brick red spots along the back. They also have a thin yellow median stripe along the length of their back.

Larva growth stages

Mature gypsy moth larva

Lifecycle

Gypsy moth has one generation per year, and includes egg, caterpillar, pupa and adult stages. Female moths lay egg masses on tree boles, branches, vehicles, houses, and other structures, and this aids their spread to new areas. Egg masses are buff-colored after they are initially deposited in late summer, but they become lighter in color as they bleach in the sun.

Adult female and male

Egg masses and pupae cases

Egg mass size may indicate population trends. When populations are declining, most egg masses are around ½ inch long and contain about 100 eggs, while building populations have 1½ inch long egg masses containing up to a thousand eggs. Gypsy moths survive the winter in the egg stage and hatch from mid-April to mid-May in Pennsylvania when temperatures are above 60 degrees Fahrenheit. During the day, they rest under leaf litter and bark crevices near the bottom of the tree. Older caterpillars are able to eat conifers, while younger stages are usually found on deciduous hosts. Mature caterpillars pupate from mid June through early July in Pennsylvania. Mice, shrews, and ground beetles eat the pupae, and are an important regulator of gypsy moth in this stage.

Adult female laying eggs

Adult gypsy moths emerge about two weeks after pupating. Adults only live about a week, and do not feed. Female gypsy moths use chemicals to attract a mate soon after they emerge. They lay eggs about a day after mating. Adult gypsy moth males have feathery antennae and brown wings and are able to fly to find females, while cream-colored females of European gypsy moths cannot fly and have threadlike antennae. There is also an Asian variety of gypsy moth with flying females that have luckily been eradicated in Western North America on several occasions following accidental introductions.

Pupae

It is fairly easy to identify gypsy moth because colors of caterpillars, adults, and egg masses are so distinct. Also, egg masses are large compared to those laid by most insects. Therefore, it is possible to predict defoliation for the following spring and prepare a suppression program to reduce the numbers of caterpillars. Caterpillars favor oak but will feed on the foliage

of many tree species, including some conifers. However, there are some trees such as ash, tulip poplar, dogwood, and black locust on which they will not feed. Defoliation by caterpillars of gypsy moth weakens trees, because without leaves the trees are not able to manufacture food. Weakened trees are susceptible to bark beetles and root diseases that can kill them.

History

Gypsy moth was introduced from Europe into Medford, Massachusetts in 1869 by Leopold Trouvelot, who was attempting to breed the insect for silk production. Unfortunately, some of the caterpillars escaped from his backyard rearing facility, and by the early 1900's they began defoliating large areas of New England.

Gypsy moth was first discovered in Pennsylvania near Pittston, in Luzerne and Lackawanna counties in 1932. During the 30's and 40's it continued to spread to Pike, Lackawanna, Wayne, Monroe, and Carbon counties. By 1969, it had spread west of the Susquehanna River, and by 1980, 38 Pennsylvania counties were infested. The gypsy moth has now moved beyond our state. It is found as far west as Minnesota, with populations reaching outbreak levels every 5-10 years.

Controls

Various natural environmental factors help control gypsy moth in North America. A disease-causing fungus known as *Entomophaga maimaiga* was first introduced in 1910-1911 to control gypsy moth. This fungus only affects select families of moth caterpillars that encounter infected soil and plants or through contact with other infected caterpillars. The spores of the fungus germinate in the spring and work best if rain is abundant. *E. maimaiga* was responsible for widespread gypsy moth mortality in 1989 and 1990, when wetter than normal conditions were reported in May. Since this time, *E. maimaiga* has become a significant regulator of gypsy moth populations at both low and high densities. Researchers are unsure whether the increased prevalence of the fungus is due to its initial introduction or if it is the result of a more recent reintroduction into the US. Older gypsy moth caterpillars that die as a result of the fungus die in a vertical position with their legs sticking outward.

Larva killed by fungus

A nucleopolyhedrosis virus (LdMNPV) kills enough gypsy moth caterpillars when populations are high to eventually end an outbreak. Caterpillars must eat the viral particles in order to become infected. Caterpillars infected with LdMNPV die in an inverted V position, which explains why the common name for LdMNPV is "the wilt". The activity of LdMNPV is specific in that it only kills gypsy moth caterpillars.

Larva killed by virus

Calosoma sycophanta

A large metallic green ground beetle known as *Calosoma sycophanta* was introduced into New England from Europe for gypsy moth control in 1906. It is now established throughout Pennsylvania. *C. sycophanta* larvae and adults eat older gypsy moth caterpillars that rest in the leaf litter during the daytime.

Other factors can impact of oak forests. Other insects, such as oak leafroller, oak leaf tier, two lined chestnut borer, and oak sawflies, as well as pathogenic fungi such as oak wilt and *Armillaria* can compound the impact of gypsy moth. In addition, high deer populations in Pennsylvania make oak regeneration a challenge.

For the latest information on gypsy moth and other forest pest consult the most recent Pennsylvania Forest Health Report posted on the DCNR-Bureau of Forestry web site.

www.dcnr.state.pa.us

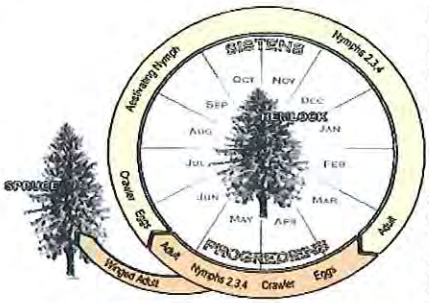
pennsylvania
DEPARTMENT OF CONSERVATION
AND NATURAL RESOURCES

Forest Health Fact Sheet
Hemlock Woolly Adelgid

Adelges tsugae, the hemlock woolly adelgid, is a fluid-feeding insect that feeds on hemlock trees throughout eastern North America, including Pennsylvania. The egg sacs of these insects look like the tips of cotton swabs clinging to the undersides of hemlock branches.

Look for hemlock wooly adelgid on the undersides of lower branches

Hemlock woolly adelgid was introduced from Asia into the Pacific Northwest in 1924. It was probably introduced into the northeastern US in the 1950's, and it was first discovered in Pennsylvania in 1967. This insect has been damaging hemlock ever since, and it is spreading. To date, 49 counties in the eastern two-thirds of PA have been infested with this insect.



Life Cycle, Courtesy of Vince D'Amico and Mike Montomery

Hemlock woolly adelgid has two generations per year in Pennsylvania. All populations are made up of females that reproduce asexually. In early spring, overwintering females lay between 100 and 300 eggs in the woolly egg sacs beneath the branches.

Mobile larvae, known as crawlers, emerge from the eggs in April or May to search for suitable feeding sites. Wind, birds and mammals often spread crawlers to nearby hemlocks. Once settled at the base of hemlock needles, crawlers become immobile nymphs which feed and mature into wingless or winged adult females by early summer. The winged form will die after searching for a suitable spruce tree that is not found in North America. The wingless form lays another 100 to 300 eggs on hemlock. Crawlers emerge from these eggs to search for suitable feeding sites. Once settled, the hemlock woolly adelgid becomes dormant until October or November, when it resumes development. Feeding continues throughout the winter and early spring.

Adelgid larvae emerge from eggs

Eastern hemlock (Pennsylvania's state tree) and Carolina hemlocks (found further south in the Smokey Mountain sections of the Appalachians) are more susceptible to hemlock woolly adelgid damage than Asian and western hemlock trees due to feeding tolerance and predators that protect the latter species. Hemlock woolly adelgid sucks fluid from the base of hemlock needles. It may also inject toxins into the tree as it feeds, accelerating needle drop and branch dieback. Although some trees die within four years, trees often persist in a weakened state for many years. Hemlocks that have been affected by hemlock woolly adelgid often have a grayish-green appearance (hemlocks naturally have a shiny, dark green color).

Other factors can influence the impact of the hemlock woolly adelgid. Other insects, such as elongate hemlock scale, hemlock borer, and spittlebugs, which are also found on hemlock, can compound the impact of hemlock woolly adelgid. Drought and fungi, such as *Fabrella* or *Korfia tsugae* can weaken hemlock and cause it to become more susceptible to insect damage. Low winter temperatures, cold snaps (episodes of freezing and thawing), and heavy thunderstorms can reduce populations of the hemlock woolly adelgid. Particularly in the mountains, it is not uncommon to find hemlocks where the insect has been killed on the top third of the trees, where it's colder and windier, but survive on the bottom two-thirds. On the other hand, mild winters can result in sharp increases in hemlock woolly adelgid populations.

PA DCNR uses integrated pest management (IPM) principles to manage hemlock woolly adelgid. IPM relies on survey and monitoring of the insect and its hemlock host. It involves using a variety of management techniques,

such as biological, chemical, cultural, and silvicultural control to reduce the populations to less damaging levels. The choice of control method will vary depending on the site and other circumstances of each situation. Unfortunately, eradication is not the objective, because hemlock woolly adelgid is already firmly established in our state.

Survey and Monitoring PA DCNR is currently attempting to map ecologically significant hemlock stands in our state, in order to detect new infestations, focus our control efforts and predict areas most vulnerable to hemlock woolly adelgid. We are using a variety of remote-sensing and ground-based techniques to accomplish this objective. Remote sensing technologies are also being evaluated for monitoring hemlock woolly adelgid. DCNR is cooperating with Rutgers to expand an algorithm developed for using Landsat imagery to detect changes in hemlock health in NJ to PA. DCNR and USFS are working with cooperators to determine if hyperspectral images taken from helicopters can detect new infestations along the leading edge and in isolated patches.

Biological control is the release of natural enemies that attack the pest. Biological controls include predators, disease organisms (fungi, bacteria, and viruses), and parasitoids (predators eat their prey, and parasitoids reproduce in their victims). Unfortunately, there are no known parasitoids of the family Adelgidae, to which hemlock woolly adelgid belongs. Pennsylvania is a key participant in national efforts to use biological control towards the management of the hemlock woolly adelgid. Unlike chemical control, biological control is a long-term, permanent strategy to managing hemlock woolly adelgid throughout Pennsylvania. In spite of these benefits, biocontrol is NOT usually recommended for landowner use. In the case of beetles, the high cost (around $3 each), large numbers of beetles required for control (thousands per site), the tendency of biocontrols to fly away from the initial release site, and the lengthy time (5-10 years) expected for noticeable results can make this method impractical. It is our hope that the biological control agents we release will, in time, protect hemlocks in ornamental situations as well as forests.

Beetles to the Rescue

Biocontrol of hemlock woolly adelgid is used in forest situations, on vigorous trees with accessible lower branches that are infested with hemlock woolly adelgid. It is likely that PA DCNR will eventually establish several natural enemies that will work together to reduce the impact of hemlock woolly adelgid.

Pseudoscymnus tsugae (PT) is a pinhead-sized, specialized, black lady beetle discovered feeding on hemlock woolly adelgid in Japan in 1992. Having completed several years of successful tests in cooperation with the U. S. Forest Service, DCNR's Bureau of Forestry has embarked on a statewide PT release effort. PT only eats hemlock wooly adelgid, balsam woolly adelgid, and pine bark adelgid. Each larva eats about 500 eggs or 50 -100 immature adelgids, known as nymphs. PT females lay up to 300 eggs in March and April, during peak egg-laying of hemlock woolly adelgid. The beetles have a second generation in June around the same time as the second adelgid generation. Adult PT feed on dormant young adelgids during the summer. Releases of PT are focused on hemlocks along the leading edge of HWA infestation. 176,387 beetles have been released at 50 sites in 23 counties. DCNR staff uses a plastic bat to beat branches over a white sheet to see if PT has established at sites in the years following release. This lady beetle should not be viewed as a cure-all, since its effectiveness is still in the testing stages. However, the data show some encouraging signs so far. To date, we have recovered 642 beetles. Adult recovery indicates overwintering success, and larval recovery indicates successful field reproduction.

Pseudoscymnus on a researcher's finger.

The lady beetles are literally grown in lady beetle farms in Pennsylvania and New Jersey, and are used for testing in hemlock sites all over the eastern seaboard. In order to make the biocontrol program more affordable, Forest Pest Management has set up a cooperative agreement with New Jersey Department of Agriculture that involves sending tons of infested hemlock foliage a year to be used for rearing hemlock woolly adelgid. In return, NJDA provides us with a substantial percentage of their PT beetles each year.

An ironic problem with the lady beetles, unfortunately, is their voracious appetite for the hemlock woolly adelgid eggs. If they are not fed enough, they tend to not multiply as quickly and can fly off in search of better feeding

grounds. However, the lady beetles have proven to be quite effective in the test sites at devouring the woolly adelgid, perhaps improving the outlook for threatened hemlocks and reducing long-term pesticide use.

More beetles

Laricobius nigrinus (LN): is a beetle that is native to the western North America where it preys on hemlock woolly adelgid on western hemlock. *Laricobius* beetles only feed on wooly adelgids. LN adults lay eggs in early spring on over wintering HWA nymphs. Larvae emerge and feed on HWA until they mature in spring, when they enter the soil to pupate. Adults remain dormant in the soil until fall. LN beetles prefer HWA to other adelgids, and it can only complete its development on this species. PA released 300 LN beetles at a site in Huntingdon County in December 2003, as part of a cooperative study with Virginia Polytechnic Institute. More releases are planned in the eastern US over the next three years. Other lady beetles are also being tested for effectiveness against hemlock woolly adelgid in Connecticut and New Jersey. A leading candidate is the *Scymnus sinuanodulus* (SS) lady beetle from China. SS is yet another predator that feeds on woolly adelgid eggs. In addition to these, Connecticut is currently working with yet another type of Chinese lady beetle, *S. ningshangiensis*. Once laboratory testing is complete, DCNR's Bureau of Forestry may add one or more of these new beetles to the hemlock woolly adelgid arsenal.

No, not that Lady beetle

Some folks who hear about DCNR's lady beetle releases are actually upset! They have had problems with another lady beetle, the **Asian Multicolored Lady Beetle**, which congregates on and in peoples' homes on sunny spring and fall days. Having the scientific name of *Harmonia axyridis* (HA), this Asian lady beetle is *huge* compared to the PT or *Scymnus* lady beetles-as big around as a pencil eraser-and is very colorful. Over one hundred PT beetles could hitch a ride on the back of one HA lady beetle! HA may be orange, brown, red, or yellow and could have black spots. It first was released by the U.S. Department of Agriculture in the 1920s to control aphids (another sucking insect) on agricultural crops. Once established, this beetle spread rapidly, until by the early '90s it could be found nearly everywhere. Attracted to warm, sheltered spots in the fall, HA prefers light-colored, sunlit houses as over wintering sites. Adults spend the winter in attics, window wells, bedrooms, and between walls until spring, when they swarm out of the houses to breed and feed for the summer.

DCNR's Bureau of Forestry gets almost as many calls each year complaining about these big lady beetles as we do about gypsy moths and hemlock woolly adelgids. Our recommendation: vacuum them up and shake the bag outside. Also, seal up areas of your home that may allow them to get inside.

Chemical control Recent research has shown that an adelgid-killing chemical injected into tree boles or applied to the ground as a soil drench or soil injection may kill hemlock woolly adelgid, and prevent new established infestations for over a year. This has prompted PA DCNR to institute a chemical suppression program on public lands at 146 sites in the state. This type of control is restricted to large, high-value (ecological, historical, or aesthetic) trees. These trees may be too tall for biocontrol releases or for application of horticultural oils or soaps. Drawbacks of chemical control include high cost of treatment, temporary control, and secondary outbreaks of spider mites. It is our hope that chemical control can be used as a "stop gap" measure to stave the hemlock woolly adelgid off and give biological control time to take effect.

Silvicultural control The DCNR is attempting to restore areas that have been impacted by the hemlock woolly adelgid. This sometimes involves replanting with native species, such as eastern white pine, that are similar ecologically, but are not affected by hemlock woolly adelgid.

Host Resistance The DCNR hopes to increase our cooperation with researchers who are attempting to identify individual eastern hemlock trees that seem to be tolerant of hemlock woolly adelgid feeding. The seed source from these individuals could be used in regeneration programs. Other researchers are currently attempting to hybridize eastern hemlock with a more tolerant or resistant host. Unfortunately attempts to hybridize eastern hemlock with three Asian species have not been successful. Also, attempts to hybridize eastern hemlock with the

morphologically similar western hemlock or mountain hemlock have been unsuccessful because these species are not well adapted to the east coast climate.

What can landowners do about Hemlock Woolly Adelgid?

Chemical Control Options: What can be done depends on the value of the trees you wish to protect. Individual ornamental trees, small trees, or even several larger hemlocks in a landscape environment can be treated with insecticides. There are several spray materials registered for application to hemlocks by ground-spraying equipment and by injection techniques. Some sprays are relatively safe to the environment, such as horticultural oils and insecticidal soaps, and others are more toxic. Oils and soaps work by suffocating the adelgid. The best time to treat is either in spring and early summer when crawlers are present, or in fall when adults break dormancy. Sprays must completely drench the needles and twigs of the entire tree to be effective, therefore this method is only recommended for trees that are 30 feet in height or less. Applications of special insecticides can be made to the tree trunk or to the soil around the tree roots. This way the tree actually moves the chemicals up to the twigs and needles where the adelgids are feeding.

Some insecticides registered for control of hemlock woolly adelgid are labeled for homeowner use. Other insecticides are restricted for use only by licensed certified pesticide applicators. Check with your county cooperative extension agent or local pest management specialist for more information, and always **read and follow the pesticide label directions**.

Both commercial spraying and injection are expensive and results with either method vary greatly depending on the quality of the equipment used, the experience of the applicator, and treatment timing (adelgid development, wind, rain, soil moisture, etc.). Call several reputable tree service companies for pricing, ask for references, and be sure to check with local clients they have served.

Forest landowners with dozens or hundreds of infested hemlocks have very few options: inject, harvest, or wait-and-see. Most Pennsylvania landowners are watching and hoping that their hemlocks survive. Some are selling their commercial hemlock timber, knowing that dead hemlock degrades very rapidly.

Tips for maintaining hemlocks and avoiding or decreasing hemlock woolly adelgid infestations:

- Do not disturb shallow roots with heavy equipment or by digging or tilling;
- Keep hemlocks well-watered (apply about 1 inch / week around drip line) during droughts;
- Do not place a bird feeder amongst your hemlock trees in infested areas of the state. Birds can transport hemlock woolly adelgid crawlers to your trees.
- Remove large, heavily infested trees that can act as reservoirs for uninfested trees.
- Clip and burn heavily infested hemlock branches. If you can catch the infestation early enough, this may significantly slow the insect's spread and build-up.
- Do not change the grade (slope of the land) near hemlocks, such as excavations or tree wells;
- Do not change water runoff patterns around hemlocks. Simply moving a downspout or installing a patio can stress these trees;
- Do NOT fertilize trees infested with hemlock woolly adelgid with nitrogen. Researchers have found five times as many hemlock woolly adelgids on nitrogen-fertilized trees, regardless of whether fertilization occurred at infestation or six months later. Once an infestation has been eradicated, fertilize hemlocks lightly with a balanced fertilizer, such as 5-10-5, late in the fall.
- When applying lime or weed killers to lawn areas, keep them at least 10 feet away from the drip line (tips of outermost branches) of hemlock trees.

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JAPANESE BEETLES IN THE URBAN LANDSCAPE

M.F. Potter, D.A. Potter, and L.H. Townsend

The Japanese beetle is probably the most devastating pest of urban landscape plants in the eastern United States. Japanese beetles were first found in this country in 1916, after being accidentally introduced into New Jersey. Until that time, this insect was known to occur only in Japan where it is not a major pest.

The eastern US provided a favorable climate, large areas of turf and pasture grass for developing grubs, hundreds of species of plants on which adults could feed, and no effective natural enemies. The beetle thrived under these conditions and has steadily expanded its geographic range north to Ontario and Minnesota, west to Iowa, Missouri and Arkansas, and south to Georgia and Alabama.

The first Japanese beetles discovered in Kentucky were found on the southern outskirts of Louisville in 1937. Isolated infestations were treated with insecticides to delay spread of the beetle. During the 1950s and 1960s, beetle populations increased dramatically and spread in Kentucky and surrounding states. Today, the Japanese beetle infests all of the counties in Kentucky.

Leaves skeletonized by Japanese beetles

Description and Habits

Adult Japanese beetles are 7/16-inch long metallic green beetles with copper-brown wing covers. A row of white tufts (spots) of hair project from under the wing covers on each side of the body.

Adults emerge from the ground and begin feeding on plants in June. Activity is most intense over a 4 to 6 week period beginning in late June, after which the beetles gradually die off. Individual beetles live about 30 to 45 days.

Japanese beetles feed on about 300 species of plants, devouring leaves, flowers, and overripe or wounded fruit. They usually feed in groups, starting at the top of a plant and working downward. The beetles are most active on warm, sunny days, and prefer plants that are in direct sunlight. A single beetle does not eat much; it is group feeding by many beetles that results in severe damage.

Adults feed on the upper surface of foliage, chewing out tissue between the veins. This gives the leaf a lacelike or skeletonized appearance. Trees that have been severely injured appear to have been scorched by fire. Japanese beetles may completely consume rose petals and leaves with delicate veins. Odors emitted from beetle-damaged leaves seem to be an important factor in the aggregation of beetles on particular food plants.

Adult Japanese beetles are highly mobile and can infest new areas from several miles away. Usually, however, they make only short flights as they move about to feed or lay eggs.

Life Cycle

Egg laying begins soon after the adults emerge from the ground and mate. Females leave plants in the afternoon, burrow 2 to 3 inches into the soil in a suitable area, and lay their eggs—a total of 40 to 60 during their life. The developing beetles spend the next 10 months in the soil as white grubs (see page 4). The grubs grow quickly and by late August are almost full-sized (about 1 inch long). Grubs feed on the roots of turfgrasses and vegetable seedlings, doing best in good quality turf in home lawns, golf courses, parks, and cemeteries. However, they can survive in almost any soil in which plants can live.

Mid-summer rainfall and adequate soil moisture are needed to keep eggs and newly-hatched grubs from drying out. Females are attracted to moist, grassy areas to lay their eggs; thus, irrigated lawns and golf courses often have high grub populations, especially during otherwise dry summers. Older grubs are relatively drought resistant and will move deeper into the soil if conditions become very dry. Japanese beetle grubs can withstand high soil moisture, so excessive rainfall or heavy watering of lawns does not bother them.

As Japanese beetle grubs chew off grass roots, they reduce the ability of grass to take up enough water to withstand the stresses of hot, dry weather. As a result, large dead patches develop in the grub-infested areas. The damaged sod is not well-anchored and can be rolled back like a carpet to expose the grubs. If the damage is allowed to develop to this stage, it may be too late to save the turf. Early recognition of the problem can prevent this destruction.

Japanese beetles overwinter in the grub stage. When the soil cools to about 60°F in the fall, the grubs begin to move deeper. Most pass the winter 2 to 6 inches below the surface, although some may go as deep as 8 to 10 inches. They become inactive when soil temperature falls to about 50°F.

When soil temperature climbs above 50°F in the spring, the grubs begin to move up into the root zone. Following a feeding period of 4-6 weeks, the grubs pupate in an earthen cell and remain there until emerging as adults.

Adult Japanese beetles

Control

From a management standpoint, it is important to recognize that both the adults and grubs can cause damage. Moreover, since Japanese beetle adults are capable of flying in from other areas, controlling one life stage will not preclude potential problems with the other. Options for protecting trees, shrubs, and flowers from adult Japanese beetles are presented below. Control of the grub stage requires properly timed applications of a soil insecticide to infested turf. Diagnosis and control of white grubs in turf is discussed in a companion publication, Kentucky Cooperative Extension Service publications ENT-10, "Controlling White Grubs" and Entfact 441, "Insecticides for Controlling of White Grub in Kentucky Turfgrass."

Plant Selection

Careful selection of plant species when replacing or adding to your landscape is the key to avoiding annual battles with Japanese beetles. Some species and cultivars are highly preferred by the adults and should be avoided where the beetle is abundant. Plants that are especially prone to damage include roses, grapes, lindens, sassafras, Norway maple, Japanese maple, purple-leaf plum, and others (Table 1). Many varieties of flowering crabapples are also severely attacked by the beetles, although some cultivars are resistant.

Roses are highly susceptible to Japanese beetles

Table 1. Landscape Plants Likely to be Attacked by Adult Japanese Beetles.

Scientific name	Common name
Acer palmatum	Japanese maple
Acer platanoides	Norway maple
Aesculus hippocastanum	Horsechestnut
Althaea rosea	Hollyhock
Betula populifolia	Gray birch
Castanea dentata	American chestnut
Hibiscus syriacus	Rose-of-Sharon, Shrub Althea
Juglans nigra	Black walnut
Malus species	Flowering crabapple, [1] apple
Platanus acerifolia	London planetree
Populus nigra italica	Lombardy poplar
Prunus species	Cherry, black cherry, plum, peach, etc.
Rosa species	Roses
Sassafras albidum	Sassafras
Sorbus americana	American mountain ash
Tilia americana	American linden[2]
Ulmus americana	American elm
Ulmus procera	English elm
Vitis species	Grape

[1]Some cultivars (e.g. Baccata v. jackii, Jewelberry, Harvest Gold, Louisa) are relatively resistant. See Kentucky Cooperative Extension Service publication ID-68, "The Flowering Crabapple," for more information.
[2]Tilia tomentosa 'Sterling' and Tilia americana 'Legend' are less susceptible than other lindens.

Fortunately, many common trees and shrubs are much less attractive to Japanese beetles (Table 2). These differences in susceptibility should be considered when selecting plant species and cultivars for use in Japanese beetle-infested areas.

Table 2. Landscape Plants Seldom Damaged by Adult Japanese Beetles.

Scientific name	Common name
Acer negundo	Boxelder*
Acer rubrum	Red maple
Acer saccharinum	Silver maple
Buxus sempervirens	Boxwood
Carya ovata	Shagbark hickory*
Cornus florida	Flowering dogwood
Diospyros virginiana	Persimmon*
Euonymus species	Euonymus (all species)
Fraxinus americana	White ash
Fraxinus pennsylvanica	Green ash
Ilex species	Holly (all species)
Juglans cinerea	Butternut*
Liriodendron tulipifera	Tuliptree
Liquidamar styraciflua	American sweetgum*
Magnolia species	Magnolia (all species)
Morus rubra	Red Mulberry
Populus alba	White poplar
Pyrus communis	Common pear*
Quercus alba	White oak*
Quercus coccinea	Scarlet oak*
Quercus rubra	Red oak*
Quercus velutina	Black oak*
Sambucus canadensis	American elder*
Syringa vulgaris	Common lilac
Most evergreen ornamentals, including Abies (fir), Juniperus, Taxus, Thuja (arbor vitae), Rhododendron, Picea (spruce), Pinus (pine) and Tsuga (hemlock) are not attacked.	

*Species marked with an asterisk may suffer occasional light feeding.

Japanese beetles are also fond of certain weeds and non-cultivated plants such as bracken, elder, multiflora rose, Indian mallow, poison ivy, smartweed, and wild grape. Elimination of these plants whenever practical destroys these continuous sources of infestation. Although plant selection is important, other approaches must obviously be used to protect susceptible plants that are already established in landscapes.

Physical Removal and Exclusion
Removing beetles by hand may provide adequate protection for small plantings, especially when beetle numbers are low. The presence of beetles on a plant attracts more beetles. Thus, by not allowing beetles to accumulate, plants will be less attractive to other beetles.

One of the easiest ways to remove Japanese beetles from small plants is to shake them off early in the morning when the insects are sluggish. The beetles may be killed by shaking them into a bucket of soapy water. Highly valued plants such as roses can be protected by covering them with cheesecloth or other fine netting during the peak of beetle activity.

Chemical Control
Many insecticides are labeled for use against adult Japanese beetles. Examples include pyrethroid products such as cyfluthrin (Tempo, Bayer Advanced Lawn & Garden Multi-Insect Killer), bifenthrin (TalstarOne, Onyx), deltamethrin (Deltagard), lambda cyhalothrin (Scimitar, Spectracide Triazicide), esfenvalerate (Ortho Bug-B-Gon Garden & Landscape Insect Killer) and permethrin (Spectracide Bug Stop Multi-Purpose Insect Control Concentrate and other brands). Carbaryl (Sevin and other brand names) too is effective. The pyrethroid products generally provide 2-3 weeks protection of plant foliage while carbaryl affords 1-2 weeks protection. For those seeking a botanical alternative, Neem products such as Azatrol or Neem-Away (Gardens Alive), or Pyola (pyrethrins in canola oil) provide about 3-4 days deterrence of Japanese beetle feeding. Insecticidal soap, extracts of garlic, hot pepper, or orange peels, and companion planting, however, are generally ineffective.
With all products, foliage and flowers should be thoroughly treated. The application may need to be repeated to prevent reinfestation during the adult flight period. Follow label directions and avoid spraying under windy conditions or when bees are foraging. Be sure the insecticide is registered for use on the plant or crop you intend to spray. If it is a food crop, note the minimum number of days that must be observed between the date of the last application and the date of harvest.

Because Japanese beetles are attracted to favored host plants from a considerable distance, controlling white

grubs in the lawn will not protect landscape plants from adult feeding.

Japanese Beetle Traps

Japanese beetle traps are sold in many garden centers. Commercially available traps attract the beetles with two types of baits. One mimics the scent of virgin female beetles and is highly attractive to males. The other bait is a sweet-smelling food-type lure that attracts both sexes. This combination of ingredients is such a powerful attractant that traps can draw in thousands of beetles in a day.

Unfortunately, research conducted at the University of Kentucky showed that the traps attract many more beetles than are actually caught. Consequently, susceptible plants along the flight path of the beetles and in the vicinity of traps are likely to suffer much more damage than if no traps are used at all.

In most landscape situations, use of Japanese beetle traps probably will do more harm than good. If you experiment with traps, be sure to place them well away from gardens and landscape plants.

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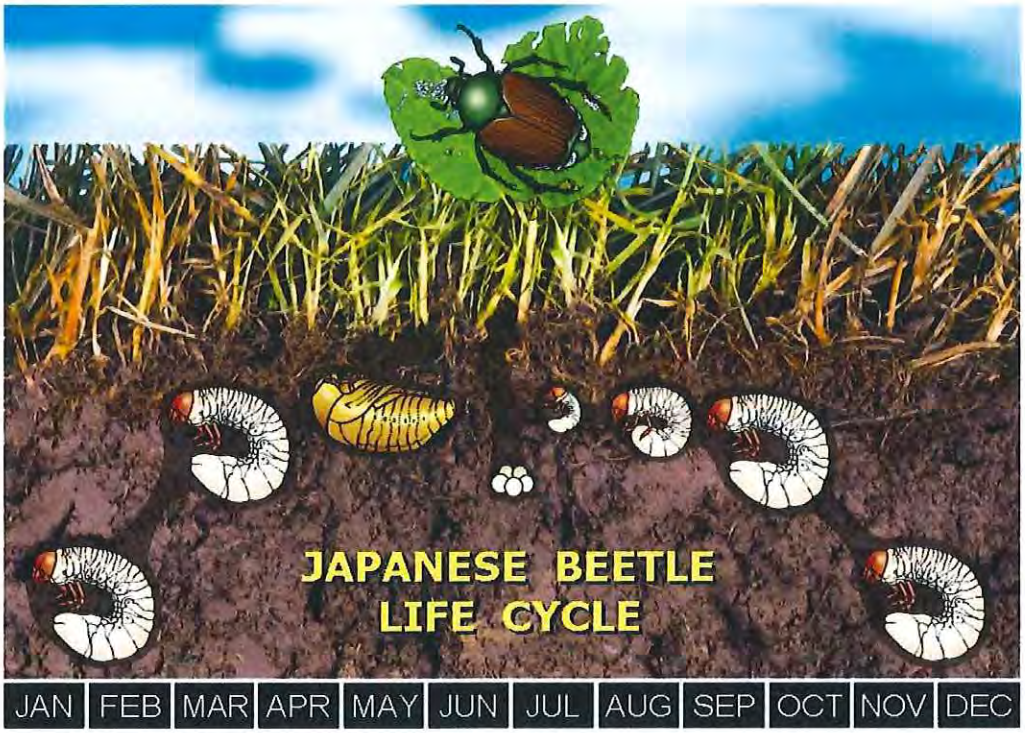

Seasonal life cycle of the Japanese beetle (courtesy of J. Kalisch, University of Nebraska)

RELATIVE SUSCEPTIBILITY OF WOODY LANDSCAPE PLANTS TO JAPANESE BEETLE *(COLEOPTERA: SCARABAEIDAE)*

By David W. Held

Abstract. The Japanese beetle (*Popillia japonica* Newman) was introduced to a New Jersey nursery in 1916 and continues to spread across the United States and Canada. Adults attack foliage, flowers, and fruit of more than 300 species of plants; however, some plants are notably resistant. This paper summarizes data on plant susceptibility of woody plants to Japanese beetles collected from observations and controlled experiments. Resistance to Japanese beetle has been documented among species of maples (*Acer*) and birch (*Betula*) and among cultivars of crabapple (*Malus*), crapemyrtle (*Lagerstroemia*), and linden (*Tilia*). Production of certain plant odors, presence of secondary compounds in leaves, and leaf pubescence are factors affecting resistance to this insect. Host plant resistance is the most sustainable means of managing feeding damage or plant losses resulting from Japanese beetle adults. When suitable, incorporating Japanese beetle–resistant plants into new landscapes can reduce or eliminate the expense of replacing damaged plants or frequent insecticide applications.

Key Words. *Popillia japonica*; Japanese beetle; integrated pest management; host plant resistance.

The Japanese beetle (*Popillia japonica* Newman) is one of the most damaging pests of urban landscapes in the eastern United States. Yearly costs for management and mitigation of damage are estimated at US$500 million (USDA/APHIS 2002). This scarab was introduced in 1916 to the eastern United States in infested nursery stock (Fleming 1972). At that time, entomologists were unaware of the pernicious nature of this species as evidenced from this quote, "While inspecting a nursery in southern New Jersey during August 1916, our attention was attracted by a scarabaeid feeding on the tips of *Crataegus*. ... Inasmuch as it was assumed to be a southern species, no particular attention to it was paid at that time" (Dickerson and Weiss 1918). In the presence of abundant grass and pasture land, and the apparent absence of natural enemies, the Japanese beetle flourished. Currently, this immigrant species partially infests or is established in all states east of the Mississippi River except Florida, and its range extends north into Canada (NAPIS 2003).

Adult Japanese beetles are broadly oval, 8 to 11 mm (about 0.5 in.) long, metallic green in color and have coppery-brown wing covers. Larvae are typical white grubs, C-shaped and cream colored, with three pairs of legs and a light-brown head capsule (Fleming 1972). Japanese beetle has a 1-year life cycle, spending most of its life underground as a grub. A small grub, about 1.5 mm (0.06 in.) long, emerges from an egg laid 3 to 5 cm (1 to 2 in.) deep into moist soil, typically under turfgrass. Females alternate between periods of feeding and mating on host plants and oviposition. During her lifetime, a female will have 12 or more egg-laying bouts and produce 40 to 60 eggs. Once hatched, grubs feed on roots and will reach full size, about 32 mm (1.25 in.) long, by late summer (Fleming 1972). Management of grubs is accomplished with soil insecticides applied preventively (e.g., Merit or Mach2) before egg hatch, or curatively (e.g., Dylox or Sevin) after small grubs are present. Presence of grubs or grub damage may be associated with nearby plants infested with adults; however, females will disperse to find a suitable site for oviposition (Fleming 1972; Potter and Held 2002).

Japanese beetle adults are active from June through August in most of its geographic range. These beetles are day active and mate and feed concurrently on host plants. They can defoliate more than 300 species of woody and herbaceous plants in 79 plant families (Fleming 1972; Potter and Held 2002). Because of their mobility and gregarious habits, swarms of Japanese beetle continually infest and defoliate new plants during the growing season. These factors can complicate control of adults, especially when using short-residual insecticides such as pyrethroids. Certain systemic products delivered through soil or trunk injection are available for control of Japanese beetles on mature, established street and residential trees; however, that treatment may not be an option for newly installed landscape plantings.

Use of insecticides to manage Japanese beetle can be reduced if resistant plants are substituted for more susceptible ones in commercial and residential landscapes. Observations and controlled experiments indicate that certain plant species, and even cultivars of the same species, vary in susceptibility. For example, moderate or complete resistance to Japanese beetle feeding is documented for most evergreens, certain crabapples (*Malus*), lindens (*Tilia*), maples (*Acer*), birch (*Betula*), and crapemyrtles (*Lagerstroemia*) (Fleming 1972; Ranney and Walgenbach

1992; Spicer et al. 1995; Potter et al. 1998; Miller and Ware 1999; Pettis et al. 2004). There is no resistance to Japanese beetle among species or cultivars of rose (*Rosa*) (Potter et al. 1998; Held and Potter 2004).

Most of the information on host susceptibility to Japanese beetle originated from a landmark survey summarized by W.E. Fleming (1972). This publication has since gone out of print; however, the information remains relevant to urban horticulture because of the continued spread of Japanese beetles into the United States and Canada. In his review, Fleming (1972) established a damage rating system based on observations of plant damage noted for each plant species in his listing. This rating system is qualitative and assigns a relative rank to each species based on written and oral accounts of Japanese beetle feeding damage noted from 1920 through 1963, primarily in the New England area (Fleming 1972).

Additional laboratory and field evaluations of Japanese beetle susceptibility for certain horticulturally important taxa have been further investigated by contemporaries of Fleming. These subsequent studies compared the percentage of defoliation of field- or container-grown plants in a common garden type of experiment (e.g., Potter et al. 1998). Blocks of woody plants representing replicates of each cultivar or species were subject to defoliation by natural beetle populations during one or more years. Additional laboratory or controlled field experiments were also used to verify the results of field tests for crabapple (Ranney and Walgenbach 1992; Spicer et al. 1995), crapemyrtle (Pettis et al. 2004), and linden (Miller and Ware 1999). Besides the field observations on *Ulmus procera*, *U. rubra*, and *U. americana* in Fleming (1972), susceptibility of elm species is based on laboratory experiments with detached leaves or defoliation of plants caged with beetles (Miller et al. 2001). Discrepancies in seasonal results from multi-year field evaluations have been noted and are attributed to the relative abundance of adults from year to year (Fleming 1972; Potter et al. 1998).

The purpose of this paper is to provide landscape architects, professional landscape managers, and arborists a comprehensive list of woody plants and their relative susceptibility to Japanese beetle. Although the results of these resistance screenings were reported in scientific or extension publications, there has been no single source of host plant data for Japanese beetle since Fleming (1972). This paper has compiled the data from Fleming (1972) and amended it with data from recent experiments to produce a comprehensive record of plant susceptibility to Japanese beetle.

DESCRIPTION OF DATA PRESENTATION

Data are presented in table form, alphabetically by family, then scientific name. Tables 1 and 4 use a rating to indicate susceptibility. This rating is an adaptation of the system used by Fleming (1972). When a plant is designated "resistant," it means observed plants were either never fed on or rarely fed on by Japanese beetles. "*" and "**" indicate plants on which feeding has been observed but is either occasional or light, respectively. "***" and "****" indicate plants that are commonly fed on by Japanese beetle, resulting in either moderate or extensive feeding damage, respectively. Plants with the latter two ratings will likely sustain considerable feeding damage or be completely defoliated if Japanese beetles are present.

Qualitative ratings for *Prunus serrulata*, *P. serotina*, and *P. virginiana* came from Fleming (1972), whereas all others were adapted from field defoliation data (Ranney and Walgenbach 1992). In the only field study with birch (*Betula*) species and cultivars, defoliation was 1% or less for all nine taxa, except for Himalayan birch (*B. jacquemontii*), which was 16% (Ranney and Walgenbach 1992). Based on these data, most birch are not preferred hosts (Table 1), except for Himalayan, European white, and gray, of which the latter two were ranked as more susceptible by Fleming (1972).

Tables 2 and 3 summarize resistance among cultivars of crapemyrtles and crabapples from field and laboratory experiments. Susceptibility ratings for crapemyrtle varieties are adapted from susceptibility rankings assigned by Pettis et al. (2004). The qualitative ratings assigned to crabapple cultivars (Table 3) were derived from three evaluations conducted in North Carolina (Ranney and Walgenbach 1992) and Kentucky (Spicer et al 1995; Potter et al. 1998). Relative susceptibility of the 26 cultivars common to both sites was similar (Potter et al. 1998).

Crapemyrtle and crabapple species or cultivars are listed under headings indicating their relative susceptibility. As before, "resistant" indicates that observed plants were rarely fed on. For crabapples, only those with less than 10% defoliation in field studies were assigned to this rating. "Moderately resistant" means that beetle feeding was observed but light. Crabapples ranked as moderately resistant generally sustained 20% to 45% defoliation. Plants designated "moderately susceptible" will have noticeable damage by Japanese beetle corresponding to 50% to 70% defoliation for crabapple varieties. All plants considered "susceptible" will be extensively damaged or completely defoliated by Japanese beetle, equivalent to about 75% to 100% defoliation in the crabapple field studies (Ranney and Walgenbach 1992; Spicer et al 1995; Potter et al. 1998).

Ratings for linden taxa (Table 4) were taken from observations in Fleming (1972), a 3-year field study of eight *Tilia* spp. in Kentucky (Potter et al. 1998), and Miller and Ware (1999), which combined laboratory feeding assays with leaves or leaf discs, with field defoliation data of 16 genotypes in Illinois. Ratings of linden were determined based on both studies; however, field defoliation data were used over laboratory results if there was any inconsistency between the relative rankings of the same variety.

Table 1. Relative susceptibility of deciduous and evergreen woody trees and shrubs to Japanese beetles.

Scientific name	Common name	Rating[z]
Aceracea		
Acer negundo	Boxelder	*
Acer palmatum	Japanese maple	****
Acer platanoides	Norway maple	****
Acer pseudoplatanus	Sycamore maple	**
Acer rubrum	Red maple	Resistant
Acer saccharinum	Silver maple	Resistant
Acer saccharum	Sugar maple	**
Anacardiaceae		
Cotinus coggygria	Smoketree	*
Rhus copallina	Flameleaf sumac	**
Rhus typhina	Staghorn sumac	*
Aquifoliaceae		
Ilex aquifolium	English holly	Resistant
Ilex cornuta	Chinese holly	Resistant
Ilex crenata	Japanese holly	Resistant
Ilex opaca	American holly	Resistant
Ilex verticillata	Winterberry holly	*
Berberidaceae		
Berberis thunbergii	Japanese barberry	**
Betulaceae		
Alnus glutinosa	Black alder	***
Betula ermanii	Erman's birch	Resistant
Betula jacquemontii	Himalayan birch	***
Betula nigra	River birch	*
Betula nigra 'Heritage'	Heritage river birch	*
Betula papyrifera	Paperbark birch	Resistant
Betula pendula	European white birch	***
Betula platyphylla var. *japonica* 'Whitespire'	Asian Whitespire birch	Resistant
Betula platyphylla var. *szechuanica*	Asian Szechuan birch	Resistant
Betula populifolia	Gray birch	****
Corylus americana	American filbert	*
Corylus colurna	Turkish filbert	**
Bignoniaceae		
Catalpa bignonioides	Southern catalpa	***
Buxaceae		
Buxus sempervirens	Common boxwood	Resistant
Calycanthaceae		
Calycanthus floridus	Carolina allspice	Resistant
Caprifoliaceae		
Lonicera fragrantissima	Winter honeysuckle	Resistant
Lonicera japonica	Japanese honeysuckle	*
Sambuscus canadensis	American elder	*
Symphoricarpos albus	Snowberry	Resistant
Symphoricarpos orbiculatus	Buckbrush	Resistant
Viburnum dentatum	Arrowwood	***
Viburnum opulus	European cranberry bush	*
Weigela florida	Weigela	**
Celastraceae		
Celastrus scandens	American bittersweet	Resistant
Euonymus alatus	Burning bush	*
Euonymus fortunei	Wintercreeper euonymus	Resistant
Clethraceae		
Clethra alnifolia	Summersweet clethra	****[y]
Cornaceae		
Cornus florida	Flowering dogwood	Resistant
Cupressaceae		
Chamaecyparis lawsoniana	Lawson white cedar	Resistant
Chamaecyparis obtuse	Hinoki cypress	Resistant
Chamaecyparis pisifera	Sawara cypress	Resistant
Chamaecyparis thyoides	Atlantic white cedar	Resistant
Juniperus chinesis	Chinese juniper	*
Juniperus communis	Common juniper	*
Thuja occidentalis	American arborvitae	*
Thuja orientalis	Oriental arborvitae	*
Ebenaceae		
Diospyros virginiana	Common persimmon	*
Ericaceae		
Kalmia latifolia	Mountain laurel	Resistant
Rhododendron catawbiense	Catawba rhododendron	*
Rhododendron maximum	Rosebay rhododendron	*
Rhododendron pericylmenoides	Pinxterbloom azalea	Resistant
Rhododendron viscosum	Swamp azalea	*
Fabaceae		
Albizia julibrissin	Mimosa	Resistant
Cercis canadensis	Eastern redbud	Resistant
Cercis chinensis	Chinese redbud	Resistant
Robinia pseudoacacia	Black locust	*
Sophora japonica	Japanese pagoda tree	*
Wisteria sinensis	Chinese wisteria	***
Fagaceae		
Castanea crenata	Japanese chestnut	**
Castanea dentate	American chestnut	****
Fagus grandifolia	American beech	**
Fagus sylvatica	European beech	**
Quercus alba	White oak	*
Quercus coccinea	Scarlet oak	*
Quercus falcata	Southern red oak	*
Quercus prinus	Chestnut oak	**
Quercus palustris	Pin oak	***
Quercus rubra	Red oak	*
Quercus stellata	Post oak	*
Quercus velutina	Black oak	*
Ginkgoaceae		
Ginkgo biloba	Maidenhair tree	*
Hamamelidaceae		
Hamamelis virginiana	Witch hazel	*
Liquidambar styraciflua	American sweetgum	*
Hippocastaneae		
Aesculus hippocastanum	Horsechestnut	****
Aesculus parviflora	Bottlebrush buckeye	***[y]
Hypericaceae		
Hypericum perforatum	Common St. Johnswort	***[y]
Juglandaceae		
Carya glabra	Pignut hickory	*
Carya ovata	Shagbark hickory	*
Carya tomentosa	Mockernut hickory	**
Juglans cinerea	Butternut	*
Juglans nigra	Black walnut	****
Lauraceae		
Lindera benzoin	Common spicebush	***
Sassafras albidum	Common sassafras	****
Loganiaceae		
Buddleia davidii	Butterfly-bush	***[y]
Buddleia alternifolia	Alternate-leaf butterfly-bush	***[y]

Table 1. Relative susceptibility of deciduous and evergreen woody trees and shrubs to Japanese beetles.

Scientific name	Common name	Rating[z]
Lythraceae		
Lagerstroemia (see Table 2)	Crapemyrtle	****[y]
Magnoliaceae		
Liriodendron tulipifera	Tulip poplar	Resistant
Magnolia grandiflora	Southern magnolia	*[y]
Magnolia × *soulangiana*	Saucer magnolia	Resistant
Magnolia virginiana	Sweetbay magnolia	Resistant
Moraceae		
Ficus carica	Common fig	Resistant
Ficus elastica	Indian rubbertree	*
Morus rubra	Red mulberry	Resistant
Musaceae		
Musa × *paradisiacal*	French plantain	*
Myricaceae		
Myrica pensylvanica	Northern bayberry	**
Nyssaceae		
Nyssa sylvatica	Tupelo	**
Oleaceae		
Forsythia × *intermedia*	Border forsythia	Resistant
Forsythia suspena var. *sieboldii*	Weeping forsythia	Resistant
Fraxinus americana	White ash	Resistant
Fraxinus pennsylvanica	Green ash	Resistant
Ligustrum ovalifolium	California privet	**
Ligustrum vulgare	Common privet	*
Syringa × *persica*	Persian lilac	Resistant
Syringa vulgaris	Common lilac	Resistant
Pinaceae		
Abies concolor	Balsam fir	Resistant
Larix deciduas	European larch	***
Picea abies	Norway spruce	Resistant
Picea orientalis	Oriental spruce	Resistant
Pinus sylvestris	Scotch pine	Resistant
Pinus virginiana	Virginia pine	*
Pseudotsuga menziesii	Douglasfir	Resistant
Tsuga canadensis	Hemlock	Resistant
Platanaceae		
Platanus × *acerifolia*	London planetree	****
Platanus occidentalis	American planetree	***
Rosaceae		
Chaenomeles japonica	Japanese flowering quince	***
Crateagus laevigata	English hawthorn	**
Crateagus monogyna	Singleseed hawthorn	**
Exochorda racemosa	Common pearlbush	**
Malus (see Table 3)		
Prunus × *cistena*	Purpleleaf sandcherry	****
Prunus sargentii	Sargent cherry	****
Prunus serotina	Black cherry	****
Prunus serrulata	Oriental cherry	***
Prunus serrulata 'Kwanzan'	Kwanzan oriental cherry	***
Prunus serrulata 'Mt. Fuji'	Mt. Fuji oriental cherry	****
Prunus serrulata 'Tai Haku'	Tai Haku oriental cherry	***
Prunus subhirtella	'Autumnalis Rosea'	****
Prunus virginiana	Common chokecherry	**
Prunus × *incamp* 'Okame'	Okame cherry	****
Prunus × *yedoensis* 'Afterglow'	Afterglow Yoshino cherry	***
Prunus × *yedoensis* 'Akebono'	Akebono Yoshino cherry	***
Pyracantha coccinea	Firethorn	*
Pseudocydonia sinensis	Chinese quince	***
Pyrus communis	Pear	*
Rosa spp. and hybrids	Roses	****[y]
Sorbus americana	American mountain ash	****
Spiraea trilobata	Three-lobed spirea	**
Spiraea × *vanhoutei*	Vanhoutte spirea	**
Rubiaceae		
Cephalanthus occidentalis	Buttonbush	***[y]
Gardenia jasminoides	Gardenia	*
Rutaceae		
Citrus sinensis	Sweet orange	***[y]
Salicaceae		
Populus alba	White poplar	Resistant
Populus alba pyramidalis	Bolleana poplar	Resistant
Populus nigra italica	Lombardy poplar	****
Salix babylonica	Babylon weeping willow	***
Salix discolor	Pussy willow	****
Saxifragaceae		
Deutzia gracilis	Deutzia	**[y]
Hydrangea arborescens	Smooth hydrangea	Resistant
Hydrangea paniculata	Panicle hydrangea	Resistant
Hydrangea petiolaris	Climbing hydrangea	*
Philadelphus coronaries	Mockorange	Resistant
Simaroubaceae		
Ailanthus altissima	Tree of Heaven	*
Staphyleaceae		
Staphylea trifolia	American bladdernut	Resistant
Styracaceae		
Halesia tetraptera	Carolina silverbell	***
Taxaceae		
Taxus baccata	English yew	Resistant
Taxus brevifolia	Western yew	Resistant
Taxus canadensis	Canada yew	Resistant
Taxus cuspidate	Japanese yew	Resistant
Taxodiaceae		
Cryptomeria japonica	Cryptomeria	*
Taxodium distichum	Baldcypress	***
Tiliaceae (see Table 4)		
Ulmaceae		
Ulmus americana	American elm	****
Ulmus changii		****
Ulmus lanceaefolia		***
Ulmus procera	English elm	****
Ulmus prunifolia		***
Ulmus pseudopropinqua		***
Ulmus rubra	Slippery elm	**
Ulmus taihangshanensis		****
Ulmus wallichiana		****
Verbenaceae		
Callicarpa dichotoma	Purple beautyberry	Resistant
Lantana camara	Lantana	Resistant

[z]Plants designated "resistant" are never fed on or rarely fed on by Japanese beetles. "*" and "**" indicate plants on which feeding has been observed but is either occasional or light, respectively. "***" and "****" indicate plants that are commonly fed on by Japanese beetle, resulting in either moderate or extensive feeding damage, respectively.

[y]Flowers of these species are also fed on by Japanese beetles.

Table 2. Relative susceptibility of crapemyrtles to Japanese beetles.

Resistant		
'Acoma'	'Pocomoke'	
Moderately resistant		
'Biloxi'	'Cordon Bleu'	'Potomac'
'Catawba'	'Lipan'	'Sioux'
'Chicksaw'	'Muskogee'	'Tuskegee'
'Choctaw'	'Osage'	'Wichita'
'Comanche'	'Pink Velour'	
Moderately susceptible		
'Apalachee'	'Hope'	'Seminole'
'Byers Standard Red'	'Hopi'	'Tonto'
'Byers Wonderful White'	'Miami'	'Tuscarora'
'Carolina Beauty'	'Natchez'	'Velma's Royal Delight'
'Centennial'	'Ozark Springs'	'Victor'
'Centennial Spirit'	'Pecos'	'William Toovey'
'Dynamite'	'Powhatan'	'World's Fair'
'Hardy Lavender'	'Raspberry Sundae'	'Yuma'
Susceptible		
'Red Rocket'	'Regal Red'	

Table 3. Relative susceptibility of crabapples to Japanese beetles.

Resistant		
Malus baccata Jackii	'Harvest Gold'	'Strawberry Parfait'
Malus hupehensis	'Jewelberry'	
'Golden Raindrops'	'Louisa'	
Moderately resistant		
'Adirondack'	*Malus halliana* var. *parkmanii*	'Red Jewel'
'Baskatong'	*Malus tschonoski*	*Malus sargentii*
'Bob White'	'Madonna'	'Sentinel'
'Brandywine'	'Molten Lava'	'Silver Moon'
'Callaway'	'Naragansett'	'Snowdrift'
'Centurion'	'Ormiston Roy'	'Sugar Tyme'
'Christmas Holly'	'Professor Sprenger'	'Wintergold'
'David'	'Profusion'	*Malus* × *zumi* 'Calocarpa'
'Doubloons'	'Ralph Shay'	*Malus* × *zumi* 'Winter'
'Gem'		
'Edna Mullins'	'Red Jade'	
Moderately susceptible		
'Adams'	'Indian Magic'	'Ruby Luster'
'Beverly'	'Indian Summer'	'Selkirk'
'Candymint Sargent'	'Mary Potter'	'Sinai Fire'
'Coralburst'	'Pink Princess'	'Snow Magic'
'Donald Wyman'	'Purple Prince'	'Tina'
Malus floribunda	'Red Baron'	'White Angel'
'Henningii'	'Robinson'	
Susceptible		
Malus baccata	'Liset'	'Royalty'
'Dolgo'	'Radiant'	'Velvet Pillar'
'Hopa'	'Red Splendor'	'Weeping Candied Apple'

Table 4. Relative susceptibility of lindens to Japanese beetles evaluated in laboratory or field experiments.

Scientific name	Common name/cultivar	Rating[z]
Tilia amurensis		**
Tilia americana		*
Tilia americana	'Legend'	**
	'Redmond'	***
Tilia caroliniana		*
Tilia chinesis		**
Tilia cordata	'Chancellor'	***
	'Fairview'	**
	'Glenleven'	**
	'Greenspire'	****
	'Olympic'	****
	'Prestige'	***
Tilia × *euchlora*		***
Tilia heterophylla	'Continental Appeal'	**
Tilia japonica		*
Tilia maximowicziana		**
Tilia mongolica		**
Tilia oliveri		*
Tilia orbicularis		***
Tilia petiolaris	Pendent silver linden	*
Tilia platyphyllos	Largeleaf linden	***
Tilia platyphyllos	'Parade'	*
Tilia tomentosa		**
Tilia tomentosa	'Erecta'	**
Tilia tomentosa	'Sterling'	**
Tilia sp.	'Sundance'	*

[z] "*" and "**" indicate plants on which feeding has been observed but is either occasional or light, respectively. "***" and "****" indicate plants that are commonly fed on by Japanese beetle, resulting in either moderate or extensive feeding damage, respectively.

DISCUSSION

Susceptibility of plants to Japanese beetle should be one factor, among many, considered when selecting plants, particularly long-lived woody plants, for residential and commercial landscapes. Resistance of one plant species to Japanese beetle does not necessarily imply resistance to other plant-feeding insects or plant pathogens (Smitley and Peterson 1993; Pettis et al. 2004). For example, the crapemyrtle varieties 'Tonto' and 'Tuscarora' are moderately susceptible to Japanese beetle, but the same varieties are resistant to metallic flea beetles (*Altica* spp.), an important pest of crapemyrtle in production (Pettis et al. 2004).

Resistance of woody host plants to Japanese beetle is probably mediated by the presence or absence of deterrent compounds found in the foliage (Keathley et al. 1999; Potter and Held 2002). Control products containing certain plant extracts, such as neem (azadirachtin), can effectively deter feeding in laboratory choice tests (Ladd et al. 1978; Held et al. 2001) but often fail to provide similar protection when tested on whole plants in the field (Harper and Potter 1994; Witt et al. 1999). Abundant field populations, however, will reduce the efficacy of both conventional and botanical insecticides because of additional adults re-infesting treated plants.

Elms and lindens are considered preferred hosts for the Japanese beetle. Among elms, only *U. lancefolia* and *U. prunifolia* were slightly less susceptible than the other species (Miller et al. 2001). Although no lindens are resistant, varieties such as 'Parade', 'Legend', and 'Sterling' appear to be less susceptible (Potter et al. 1998). Moderate to dense leaf pubescence may be an important factor in susceptibility of linden and elm to Japanese beetle. For example, foliage of *T. platyphyllos* 'Parade', *T. tomentosa* 'Sterling', and *U. lamellose* have heavy pubescence and is less preferred by Japanese beetle (Potter et al. 1998; Miller and Ware 1999; Miller et al. 2001). Conversely, certain plants, such as species of *Ilex* and *Rhododendron*, with waxy or glossy foliage are also resistant to Japanese beetle (Fleming 1972; Keathley et al. 1999).

Plants with purplish or deep red foliage (e.g., 'Crimson King' Norway maple) are often observed to sustain more damage by Japanese beetle than green-leaved cultivars (Rowe et al. 2002). Foliage color alone, however, does not account for these differences. When two artificial ficus trees with foliage painted either green or purple are placed side by side in the field, significantly more beetles land on the green-leaved plants (Rowe et al. 2002). Flower color, however, does influence susceptibility of flowering plants to Japanese beetle. Rose varieties with yellow or white flowers are more likely to be attacked than those with darker-colored blooms (Held and Potter 2004).

Resistance of certain plants to Japanese beetle also depends on the production of attractive volatile compounds following damage by Japanese beetle or other plant-feeding insects (Loughrin et al. 1995). Japanese beetles use a wide range of floral and fruitlike compounds to locate a host plant (Fleming 1972; Loughrin et al. 1995, 1996). Laboratory tests show that Japanese beetle often cannot discriminate among foliage of plants that differ in susceptibility in the field (Loughrin et al. 1995, 1996). This finding indicates that Japanese beetles are attracted to plants regardless of their status as a host (Potter and Held 2002). However, if susceptible plants suffer feeding damage, they produce an array of attractive volatiles that serve as aggregation stimulants for Japanese beetle (Loughrin et al. 1995, 1996). Therefore, a susceptible plant in the field may not be inherently more attractive, but, if damaged, these plants produce the volatiles that recruit Japanese beetles much like sharks attracted to a blood trail in the water.

Host plant resistance is the most sustainable means of managing feeding damage or plant losses resulting from Japanese beetle adults. Landscape designers in states on the front of this insect's range expansion should consider incorporating resistant plants into residential, commercial, and municipal landscapes as well as any other long-term

plantings. This approach can reduce the economic and environmental costs associated with the repeated use of insecticides to prevent or reduce damage to urban landscapes in the future.

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Mountain Ash Sawfly

Pristiphora geniculata (Hartig); Family: Tenthredinidae

Young larvae feeding gregariously.
Photo from www.forestryimages.org
Steven Katovich, USDA Forest Service.

Mature larva.
Photo from
www.forestryimages.org
James B. Hanson,
USDA Forest Service.

Injury

The mountain ash sawfly can defoliate a tree in a short time when it is present in large numbers. The European and American mountain ash (*Sorbus aucuparia* and *S. americana*) are the known hosts. Damage can be seen in late June or early July. The larvae feed on the leaves and devour all but the mid-ribs and larger veins.

Description

The larval stage of this wasp causes the damage. At first glance, sawfly larvae resemble caterpillars of moths or butterflies, but sawflies have more pairs of leglike appendages. The young mountain ash sawfly larvae are greenish with black dots down the sides, and with black legs and a head. As they reach maturity, the head and legs become distinctively yellow-orange. The larvae feed gregariously.

The adult sawfly is a stout-shaped wasp, and is yellow with black spots. Pupae are tan to brown, oval in shape, and are found on the ground under the tree.

Life History

In late May or early June adult sawflies emerge from the overwintering cocoons, and the females begin to lay eggs in slits cut into the leaf edges. The tiny larvae begin feeding as soon as they hatch from the eggs, and they increase gradually in size over the next three to four weeks. When the larvae are fully grown, they drop to the ground and spin cocoons. There is usually one generation per year, but at times, a partial second generation may occur. The larvae feed in groups starting on one or two branches, but soon moving on to others as the food source is depleted.

Management

For small trees, remove larvae by hand, by clipping or pulling off infested leaves.

Where this is not possible, insecticides may be applied as soon as the first larvae are seen, usually in early to mid-June (448-707 GDD*), PPI (Plant Phenology Index) when cranberry bush or mock orange are flowering. If you had damage this past year, make a note on your calendar to check the trees in June and July of the coming year for the presence of larvae. Scout weekly in July and August for the possibility of second generation, and if needed treat when larvae appear.

*GDD = Growing Degree Days (Base 50°F). Your local radio station may make this information available, or see this website: http://www.nrcc.cornell.edu/grass/degreedays/degreedays.html

Prepared 1983 by Carolyn Klass, Senior Extension Associate, Dept. of Entomology, Cornell University
Updated 2012

THE OHIO STATE UNIVERSITY

BUGDOC

Christmas Tree

Fact Sheet

Pine Bark Adelgid

Pine bark adelgids can nearly coat the surface of pines.

Pine bark adelgid females lay masses of eggs in cottony wax threads.

Species: *Pineus strobi* (Hartig)

Distribution: North America and Europe

Hosts:

Mainly attacks eastern white pine but can infest Scotch, Ponderosa, jack and pitch pines.

Damage:

Heavy infestations on branches of pines will cause stunted growth. Foliage may become blackened from a fungus growing on the honeydew and wax produced by the nymphs and adults. Occasionally even tree death may occur. This pest usually does not attack smaller trees.

Description and Life Cycle:

This pest is often called an aphid though adelgids do not have the long antennae and cornicles typical of aphids. The life cycle of the pine bark adelgid is not well understood. Apparently, immature females overwinter attached to the bark of pines. When the spring temperatures reach 50F, these nymphal females become active and are soon covered with a white woolly wax. By late April, the nymphs mature and molt into wingless adults. These females lay 40 to 50 eggs in 20 to 30 days. The eggs hatch in one to two weeks and the crawlers move to suitable places on the tree to insert their mouthparts. They often hide under old dead adults on the bark, at the bases of old needles and under newly emerging needles. The new nymphs produce waxy coverings and take about 20 to 30 days to mature. This pest may have up to five overlapping generations in a season with adults, nymphs and crawlers present throughout the summer. Adults and young nymphs apparently die during the winter; only the nymphs which have molted twice, third instars, can overwinter.

Control Hints:

This pest is slow to spread because the crawlers have to drop from adjacent trees, be wind

blown, or hitch a ride on birds or other animals found in the plantation. Older stands of white pines surrounding plantations should be removed.

Option 1: Biological Control - Encourage Natural Predators and Parasites - Many lady beetles, lacewings and hover flies attack the pine bark adelgid. These are usually not effective in Christmas tree plantations because of the use of insecticides sprayed for other pests. Be careful to not misidentify the lady beetle larvae which are often covered with white woolly material and look much like the adelgids.

Option 2: Chemical Control - Dormant Oil Sprays - Dormant oil sprays (3-5%) are quite effective if applied in the fall or spring to kill the overwintering nymphs. Be sure to spray in the spring before the females have begun to produce eggs in the waxy coating. Thorough coverage of the trunk and branches is needed.

Option 3: Chemical Control - Insecticide Sprays - Sprays using registered insecticides, applied in mid-April will kill the overwintering nymphs before they mature and lay eggs. Summer sprays are effective but two to three sprays, at weekly intervals, will be needed to kill new crawlers hatching from resistant eggs.

Revised 05/2002; © D.J. Shetlar

PRUNING TO MANAGE WHITE PINE WEEVIL

Insect and Disease Laboratory • 50 Hospital Street • Augusta, Maine • 04330-6598

Background

The white pine weevil, which kills the tops of conifers, is the most serious economic insect pest of white pine in Maine. Weevil attacks result in trees that are multi-topped, crooked, and of much lower value for sawtimber. Appearance and development of ornamental trees are also affected. Trees in open areas, plantations, and forest clearings are most severely damaged.

White pine weevil most commonly attacks eastern white pine, jack pine, and Norway spruce. However, many other pines and spruces, including ornamentals, are also susceptible.

Symptoms

Figure 1. Pitch from adult feeding punctures

One of the first symptoms of attack in the spring is the presence of pitch flowing from feeding punctures in the previous year's leading shoot (figure 1). Beginning in late June and early July, the new growth on infested shoots starts to droop (figure 2). Shortly thereafter the tops die and turn brown. Up to 2-3 years of top growth may be destroyed (figure 3).

Figure 2. Early signs of wilt.

Figure 3. Damage seen later in the summer (photo: E. Walker, MN Dept. of Nat. Res. Archives, Bugwood.org)

Life Cycle

Starting in early spring, weevil eggs are laid in the bark of the previous year's leader. After hatching, the legless grubs start tunneling downward under the bark, eventually girdling the infested stem and killing the top 1 1/2 - 2 feet of the tree. Larvae pupate in the main stem, and by mid-July adults begin to emerge. **To have any effect, pruning must be done before this time.**

Pruning of Infested Leaders - Pruning and destroying infested terminal shoots on young trees in late June and early July kills the current year's generation of beetles, and can reduce attack in future years (figure 4).

- **PRUNE** infested leaders (the topmost shoot) at a point just into healthy tissue below the tunneling grubs.
- **DESTROY** by promptly burning all cut leaders to kill the insects before adults emerge. Do not leave removed material on site, since weevils will develop and emerge from cut material.

Figure 4. Early summer damage. Prune at arrow.

WHEN TO PRUNE: Wilting of leaders may be detected anytime after late June. Pruning should be completed when the wild raspberries start to ripen (early to mid-July, depending on your location), while weevils are still in the wilted or dead terminal shoot. Pruning before completion of shoot growth encourages secondary budding on the pruned leader and may result in faster terminal recovery. Early pruning can also remove the weevils when only one season's growth has been damaged, rather than two or three. **Pruning after mid-July, once the adults have emerged, is not effective.**

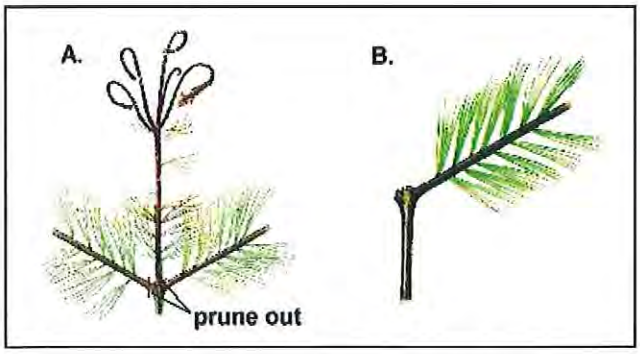

Figure 5. Pruning following a weevil attack should remove the wilted or dead terminal and all laterals except one. This lateral will straighten over a period of a few years (illustration: Julie Janke, Scientific Illustrator, Afton, MN).

Corrective Pruning – After the removal of infested leaders, tops of trees should be pruned in such a manner that all but a single dominant shoot at the topmost healthy whorl are removed to encourage formation of a new leader (figure 5). Corrective pruning may be postponed until the following year to make sure that at least one live lateral will survive possible damage (e.g. from ice, snow, or a second year of weevil attack).

Shade – Partial shade helps reduce weevil attack by reducing temperature and bark thickness. Leaving some overtopping hardwoods or other conifers may help protect young pine and spruce from weevil damage when they are most susceptible. Density of the overtopping species should be managed to maintain about 45-50% shade until final release when the trees reach a height of 16 to 18 feet. Closer spacing of trees in a plantation will stimulate height growth and natural pruning to permit faster recovery.

Pest Alert

United States
Department of Agriculture
Forest Service
Northeastern Area
State and Private Forestry
NA–PR–01–12
November 2012

Winter Moth

Winter moth, *Operophtera brumata* L. (Lepidoptera: Geometridae), is a non-native invasive defoliator from Europe that was discovered in Massachusetts in the late 1990s. Winter moth has now been found throughout the eastern half of Massachusetts and into Rhode Island, Connecticut, Long Island (NY), southeastern New Hampshire, and southeastern Maine. It is expected to continue to spread to suitable habitats.

Winter moth has been responsible for widespread and prolonged defoliation in eastern Massachusetts. Winter moth is of particular concern because it feeds on many species and is a pest of deciduous forests, fruit orchards, and ornamental trees and shrubs in its native and exotic ranges. High densities of winter moth can completely defoliate hardwood stands and cause trees to refoliate in the late spring and/or early summer, which may further reduce tree nutrient stores.

Winter moth defoliation has reduced radial growth of oak trees in Massachusetts and has been reported as the causal agent of tree mortality in some stands. Given the early-spring feeding of winter moth and the existence of a well-developed native and exotic defoliator complex, hardwood forests of New England may be subject to multiple defoliators in a single season. Defoliation by winter moth may initiate tree decline that involves secondary pests and decay fungi.

Hosts

Winter moth has a wide host range. In North America, high levels of defoliation have been noted on oak (*Quercus*), maple (*Acer*), birch (*Betula*), apple (*Malus*), and blueberry (*Vaccinium*) species. Winter moth also feeds on ash (*Fraxinus*), basswood (*Tilia*), hazelnut (*Corylus*), beech (*Fagus*), elm (*Ulmus*), and many other genera. In Europe and British Columbia, winter moth also feeds on spruce (*Picea*). Feeding damage largely depends on synchrony between winter moth egg hatch and host tree bud break.

Description

Winter moth is similar in appearance and life history to native insects including fall cankerworm (*Alsophila pometaria*) and Bruce spanworm (*Operophtera bruceata*).

Distinguishing winter moth from the native Bruce spanworm is difficult where the two species overlap and typically requires DNA analysis.

Winter moth produces small (0.42 - 0.70 mm), light green, ovoid eggs that change to orange and then dark blue prior to hatching. Winter moth larvae are lime green "inchworm" ("looper") caterpillars with two pairs of ventral prolegs and faint white to creamy-yellow stripes running lengthwise along each side of the body (figure 1). The brown pupa is about 7.0 to 7.6 mm long and features two short spines at the rear (figure 2).

Figure 1.—Winter moth larva.

Figure 2.—Winter moth pupa.

The adult male moth is tan to brown with a wingspan up to 30.0 mm and is capable of flight. The male forewings are fringed and feature bands of black hatch marks (figure 3). The female moth is grayish black and flightless. The vestigial forewings of the female moth are 2.0 to 3.2 mm long and cover the first two to three abdominal segments (figure 4). Winter moth females have longer vestigial forewings than Bruce spanworm females, making this the easiest life stage to distinguish between the two species.

Figure 3.—Adult male winter moth.

Figure 4.—Adult female winter moth.

Life History

In its native European range, winter moth populations are cyclical, with 2- to 3-year outbreaks occurring approximately every 8 to 10 years. In Massachusetts, high densities of winter moth have been consistently observed

for more than a decade. Winter moth feeds at the same time as fall cankerworm and Bruce spanworm, and can hybridize with the latter. In eastern Massachusetts, winter moth has now become the dominant early-spring defoliator.

Winter moth completes one generation per year and overwinters in the egg stage. Eggs typically hatch in April and larvae begin feeding within the expanding buds and later on the foliage for approximately 6 weeks. Winter moth feeding can completely strip trees of foliage (figure 5, left), leaving only the veins. Lighter levels of defoliation can produce tattered leaves with a sieve-like appearance (figure 5, right). Much of the damage occurs inside the bud before the leaves expand. Winter moth early instar larvae disperse in the wind from tree crowns via silken threads in a process known as "ballooning."

Figure 5.—Winter moth feeding damage: heavy (left), lighter (right).

Beginning in mid- to late-May, larvae stop feeding and drop from tree crowns to the soil to pupate. The pupae form inside cocoons made of silk mixed with soil in the litter beneath trees where larvae fed. Adults emerge from the soil in late fall to early winter, with males dispersing in search of mates. After mating, the flightless females crawl up tree boles and lay eggs singly or in clusters of two to three in bark crevices, on or under lichen, and around the buds of tree branches. A female winter moth produces an average of approximately 150 eggs. Adults are active in the evening, and the male moth can often be seen fluttering around lights, even on cold nights. Because winter moth has a limited potential to disperse naturally, it is likely introduced into new areas through its inadvertent movement in plant material (firewood, ornamental plants, and nursery stock containing soil).

Management and Biological Control

An integrated pest management approach will be needed to manage winter moth where it occurs. It is important to maintain "vigorous" trees and stands through appropriate silvicultural techniques that reduce stress and damage to residual trees. Insecticidal control may be helpful in reducing winter moth populations under certain circumstances, but timing applications with tree phenology is important. Insecticides are only effective during the larval stages and it is critical that application follows host tree bud burst, as early instar larvae are protected when feeding within tree buds.

Currently, biological control may be the most practical tool to manage winter moth populations in forest stands or at larger scales. Previous winter moth invasions in Nova Scotia and British Columbia have been successfully controlled by introducing *Cyzenis albicans*, a parasitoid fly that feeds exclusively on winter moth. Efforts to establish *C. albicans* to manage the burgeoning winter moth population in Massachusetts began in 2005. To date, small populations of this biological control agent have become established at six release sites, and the percent parasitism of winter moth is increasing at some of these sites. Given the high density of winter moth populations throughout eastern Massachusetts, continued release of *C. albicans* is needed to manage this pest.

Photographs:

Figure 1: Milan Zubrik, Forest Research Institute - Slovakia, Bugwood.org

Figure 2: Hannes Lemme, Bugwood.org

Figure 3: Molly Heuss, USDA Forest Service

Figure 4: Molly Heuss, USDA Forest Service

Figure 5 (left): Thomas Lee, University of New Hampshire

Figure 5 (right): Michael Simmons, USDA Forest Service

Pesticide Precautionary Statement

Pesticides used improperly can be injurious to humans, animals, and plants. Follow the directions and heed all precautions on the labels. NOTE: Some States have restrictions on the use of certain pesticides. Check your State and local regulations. Also, because registrations of pesticides are under constant review by the Federal Environmental Protection Agency, consult your county agricultural agent or State extension specialist to be sure the intended use is still registered.

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Anthracnose of Maple

[1]

[2]

Anthracnose of maple is often a minor disease that only reduces the aesthetic value of an infected tree. Under ideal conditions, however, the disease can be severe, leading to premature defoliation and contributing to decline when other diseases and/or insect pests are present. Healthy trees may undergo defoliation in spring shortly after leaf out but are able to flush a new set of foliage and recover. Young trees are more susceptible to lasting damage while older, established trees can usually survive multiple years of defoliation.

Pest

Anthracnose of maple is caused by several fungi including *Aureobasidium apocryptum* (syn. *Gloeosporium apocryptum* and *Kabatiella apocrypta*), *Discula campestris* (syn. *Gloeosporium campestre*), *D. umbrinella* (syn. *Gloeosporium umbrinellum*) and *Colletotrichum* spp. (syn. *Glomerella* spp.)

Hosts

Maple (*Acer*) species including sugar (*A. saccharum*), red (*A. rubrum*), Norway (*A. platanoides*), silver (*A. saccharinum*) and Japanese (*A. palmatum*) maples.

Symptoms & Disease Cycle

Symptoms vary by host and by the fungal pathogen present but are characterized by irregularly-shaped, angular spots or blotches that occur primarily along leaf veins or margins **(Figure 1)**. Lesions on Norway maple tend to be narrow, purplish-black streaks along leaf veins. On sugar maple, lesions occur between leaf veins and appear as large, brown blotches. On Japanese maple, lesions occur between veins or at leaf margins as light-brown to tan, papery spots. Anthracnose fungi overwinter within senescent leaf tissue on the ground and in infected twigs and buds. Spores produced on this infected tissue in the spring are spread by wind and splashing rainwater. Anthracnose fungi produce asexual spores (conidia) in acervuli which appear as dark-brown to black spots within lesions. Acervuli can be found on the upper or lower leaf surface and along veins or midribs **(Figure 2)**. Spores are produced whenever environmental conditions allow (mild and wet) from spring through late-summer. Severe infections cause premature defoliation and may cause distortion of young leaves. Symptoms on twigs and buds are less common on maples but under high disease pressure, young shoots may be killed.

Management

.ıfected leaves shed by the tree are the primary source of inoculum and should be removed from the site. If the disease is severe, infected shoots can be pruned out, as they harbor overwintering spores. This sanitation practice will reduce the amount of spores present to re-infect trees during the spring and throughout the next growing season. Maintaining tree vigor through adequate fertilization, watering, mulching and pruning will help lessen the impacts of disease on tree health. Sugar maple prefers calcareous soils and many

respond well to lime applications if soil pH is too low. Anthracnose is often an aesthetic issue on maple, but there are situations where the disease can cause permanent damage and chemical control may be warranted. These situations include when: young trees are heavily infected; trees are severely defoliated for consecutive years, and; trees are in decline due to environmental stress or attack by insect pests or other disease-causing organisms. If necessary, labeled fungicides can be used to protect at-risk trees. The following products are labeled for use against anthracnose of maple: chlorothalonil, chlorothalonil + thiophanate-methyl, copper hydroxide + mancozeb, mancozeb and propiconazole. The first application should be made at or just before bud break to protect new growth from initial infection in spring. Additional applications may be required at the labeled interval, especially if prolonged periods of wetness occur.

Author: Nicholas
Last Updated: Apr 29, 2015
Topics: Commercial Horticulture
Commercial Horticulture topics: Diseases

Source URL: https://ag.umass.edu/fact-sheets/anthracnose-of-maple

Links:
[1] https://ag.umass.edu/sites/ag.umass.edu/files/fact-sheets/images/maple_anthracnose_figure1.jpg
[2] https://ag.umass.edu/sites/ag.umass.edu/files/fact-sheets/images/maple_anthracnose_figure2.jpg

Cornell University
College of Agriculture and Life Sciences

Plant Disease Diagnostic Clinic
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Black Knot: *Apiosporina morbosa*

Introduction

Black knot disease occurs on numerous cultivated and wild plums, prunes, and cherries (*Prunus* spp.). The disease is characterized by the presence of warty, black galls which may vary in size from 1/2 inch to more that 1 foot in length. In some parts of the Northeast and Midwest, black knot causes serious losses to commercial plum and prune growers. More often, however, the grotesque galls draw the attention from homeowners who want to improve the unsightly appearance of affected landscape trees.

Figure 1: Branches appear thick and black. (provided by **Dr. Phil A. Arneson, Cornell University**)

Black knot appears to be a minor problem on *Prunus* spp. found in forest situations, where susceptible trees are surrounded by many non-susceptible species of trees. Black Knot is mainly a problem in North America (Canada, the United States and Mexico) where it is indigenous. A record from 1979 indicated the presence of the fungus on pear in Taiwan but no other incidences have been reported from Asia. Currently it is not found in Europe or the EPPO region. The EPPO region consists of 43 European and Mediterranean countries that are responsible for international plant protection in their region. *Apiosporina morbosa* (syn.= *Dibotryon morbosum*) is listed as an EPPO A1 quarantine pest. Inclusion on the list requires the countries to follow phyto-sanitary regulations and to make appropriate requirements for admission of plant material in their countries.

Black knot has been reported on 24 *Prunus* spp. but is most commonly found on wild and cultivated plums and cherries. Early publications from the 1950's describing the disease report no cases affecting peach, but a few rare infections have been reported since that time. The disease can be found throughout North America but is most commonly found in the northeast. It was first reported as a destructive disease in Massachusetts in 1811. It was first described in 1821 by L. D. Schweinitz from specimens collected in Pennsylvania. Researchers believe the disease is caused by a native pathogen that was only found in the northeastern states until around 1875, when observations of the disease began arising in the central states.

Young, infected twigs may die during the first year of infection. Larger branches may take several years to display severe damage. The infected trees decline and become more symptomatic with each growing season. The infection stresses the entire tree causing it to weaken, decline, and possibly die. The stress placed on the tree may also make it susceptible to infections by other pathogens. Economically, the trees lose value after a few years. The portions of a branch distal to a knot become stunted, and occasionally knots enlarge to girdle a branch and possibly kill it. Trees with multiple infections become dwarfed and misshapen, markedly reducing their productivity and attractiveness.

Because of the long infection process and disease cycle, this disease is often overlooked by home gardeners and fruit producers. The leaves can mask the symptoms until firmly established infections are in place. Once established, it is very difficult to manage the disease. Commercial growers often discover the disease more quickly because they regularly inspect their trees during routine crop management. Awareness and strict monitoring of susceptible plants should be a priority for all home gardeners and commercial growers.

Symptoms and Signs

The disease is characterized by the presence of thick, black, irregular swellings on the twigs (**Fig. 1**). The presence of these symptoms is often first noticed in the winter season when they are not obscured by leaves. However, the fungal disease-causing agent has been present for quite some time. The pathogen's presence disrupts the normal growth of the twigs and a tumor-like growth forms at the infection site. Infections may take place as much as a year or more prior to the development of these characteristic "knots", therefore, the swellings are normally not noticed until the winter of the second season of infection. It takes a keen observer to notice the subtle, initial symptoms present during the first season of infection.

Figure 2: Galls caused by the fungus on cherry. (provided by Dr. Phil A. Arneson, Cornell University)

The first symptoms appear as small, light brown swellings of the current or previous season's growth. By the next season the swellings turn olive-green in color with a velvety texture. Over this growing season the knots darken and appear to have a hard, brittle texture (**Fig. 2**). The hard, black knots are the typical symptoms associated with the disease.

Knots vary in size from approximately 1-30 cm (0.5 to 12 in.) in length and from minute measurements to 5 cm (2 in.) in circumference. The infected twigs often appear bent at the tips because of extra cellular growth on one side. Trees with heavy infections may contain numerous knots. Some of the older knots may appear white or pink in color. This discoloration is often seen in late summer and is caused by the fungal parasite, *Trichothecium roseum.*

Disease Cycle

The fungus overwinters in the knots. About the time of bud emergence in the spring, the first ascospores are forcibly discharged from the ascostromata following a period of warm, wet weather. Apparently very short periods of wetness (only a few hours) are enough to prompt ascospore discharge. Temperatures between 16 and 27°C (60-80°F) are ideal for the dissemination, germination, and infection of new plant tissue. Recent studies have confirmed and concluded that rainfall and temperature are the key factors in the release of spores and that the duration of the rainfall or wet period is not a factor.

The ascospores are spread by air currents and rain splashing. Mainly the succulent green shoots and, occasionally, wounded tissues are most susceptible to infection by ascospores. Ascospore discharge continues to occur for 2-3 weeks after bloom. Infections take place during this time but may continue for a longer time period if susceptible host plant tissue is available. The germinating ascospores have the ability to penetrate unwounded surfaces of elongating, green shoots directly.

The knots develop very slowly, and by the end of the summer they appear only as small galls that might easily be overlooked (**Fig. 3**). Further development does not occur until the following spring when the knots enlarge very rapidly. They initially are quite soft in texture and become greenish-brown in color as conidia develop over their surfaces (**Fig. 4**). The

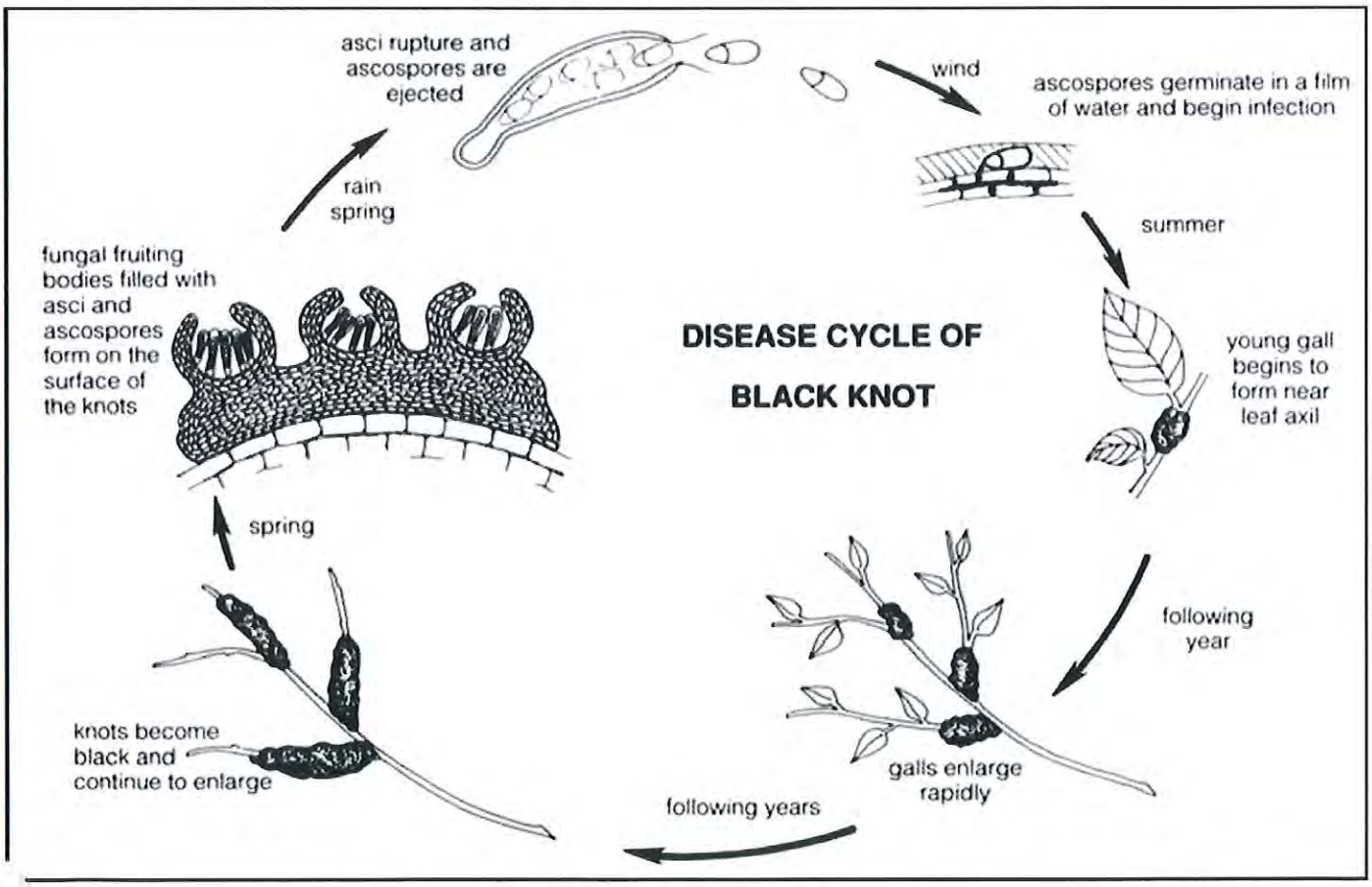
Figure 3. The early stages of knot formation result in only small galls. (provided by **Dr. Phil A. Arneson, Cornell University)**

The conidia are disseminated by wind and splashing rain but probably do not figure as prominently as the ascospores in establishing new infections. By the second summer after infection, the knots have enlarged considerably and begin to change to a hard, coal-black structure. The old knots enlarge every year by advancing at the margins. The fungus mycelium can also spread internally and give rise to new galls some distance from the original knot. The central, older portions of the knot eventually break down and are invaded by boring insects.

Management Strategies

Cultural management

Cultural management strategies are important in black knot management. Sites containing *Prunus* spp. should be monitored on a scheduled basis for possible infections. The main strategy to lower disease incidence is the removal of sources of inoculum. All shoots and branches bearing knots should be pruned out during the winter. This pruning should be completed before ascospore discharge begins in the spring, usually about the time that the buds first break. To be sure that even the unseen internal mycelium is removed, the cut should be made at least 15-20cm (6-8 in.) below the knot. Winter is also a



Black knot disease cycle.
(provided by **Dr. Wayne Wilcox, Cornell University, NYSAES, Geneva, NY)**

good time to look for and remove sources of inoculum in nearby wild *Prunus* spp. in hedgerows and woodlots. The knots are capable of producing ascospores for some time after removal from the tree. Therefore, they should be burned, buried, or removed from the site regardless of the time of year the pruning takes place.

Figure 4. As knots develop, they become very soft in texture and are covered with olive-green conidia. (provided by **Dr. Phil A. Arneson, Cornell University)**

Genetic resistance

When selecting trees for a new planting, consider selecting varieties with known resistance. Varieties may vary in their ability to tolerate or resist an infection. Black knot resistance is as important as fruit characteristics, tree size, and flowering time. Up-to-date listings of varieties with high levels of resistance are often available from local extension offices. The plum variety President has shown high resistance. Moderately resistant varieties include: Methley, Milton, Early Italian, Brodshaw, Fellenberg, Shiro, Santa Rose, and Formosa. Shropshire and Stanley are very susceptible.

Site selection

Consider the site location. Avoid planting new trees near areas with known problems such as abandoned orchards or where wild varieties have been observed with the disease. When possible, remove wild varieties of the trees from the area.

Biological control

Interest in biological control agents is increasing because of the loss of certain fungicide registrations and the fact that applicators would prefer to reduce their exposure to pesticides. A possible biological control agent for black knot may be the fungal parasite, *Trichothecium roseum*, introduced in the symptoms section.

Chemical management

Fungicides to manage black knot are normally only recommended for commercial fruit production in New York State. At this time, there are no products recommended in New York for use on home orchard fruit trees or ornamentals for managing Black Knot. Homeowners should practice cultural controls for both ornamental and home orchard fruit trees that show evidence of this disease.

Updated SLJ 1/15

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CEDAR-APPLE RUST

Cedar-apple rust is a distinctive disease that is indigenous and widespread throughout the Northeast in areas where apples and crabapples grow in close proximity to members of the juniper family.

SYMPTOMATOLOGY AND DISEASE CYCLE:

Cedar-apple rust, caused by the fungus *Gymnosporangium juniperi-virginianae*, is a heteroecious rust that requires two different hosts to complete its two-year life cycle. The primary hosts are species of *Malus*--apple (*M. domestica*) and crabapple (*M. sylvestris* and other *Malus* species). The alternate hosts are members of the genus *Juniperus*, which includes the native Eastern red cedar (*J. virginiana*) as well as many ornamental junipers. The fungus must spend part of its life cycle on both hosts. The cedar-apple rust fungus is a *Gymnosporangium* rust and has a similar life cycle and is closely related to several other *Gymnosporangium* rusts, including cedar-hawthorn rust and cedar-quince rust.

The symptoms of cedar-apple rust disease on Eastern red cedar and other junipers are inconspicuous during the winter and appear as brown, kidney-shaped galls that vary in size from ¼-2 inches in diameter (Figure 1). As the temperatures begin to rise in the spring, the fungus begins to grow in the galls (Figure 2).

Figure 1. Dormant cedar-apple rust gall overwintering on Eastern red cedar.

Figure 2. Gall with telial horns beginning to emerge in early to mid-spring.

After periods of cool, rainy weather, the spectacular and distinctive bright orange, gelatinous spore horns develop and protrude from the surface of these galls (Figure 3). They can be up to four inches long. Heavily infected junipers appear to be "decorated" with many colorful galls (Figure 4).

Figure 3. Spectacular, gelatinous telial horns develop from galls after rain.

Figure 4. Eastern red cedar "decorated" with many galls in spring.

Spores called teliospores are produced in these gelatinous spore horns or tendrils. As the teliospores germinate, they produce another type of tiny spore, called a basidiospore. These spores can only infect apple and crabapple. Basidiospores are released and carried by wind and driving rain to newly emerging leaves of the alternate hosts, apple and crabapple. As many as 7.5 million basidiospores may be produced in a single gall. These spores have been shown to be carried as far as six miles. Once the spores land on the emerging apple or crabapple leaves, they germinate and infect the leaves when they are wet.

Symptoms of infection on the apple and crabapple hosts are also quite colorful. Lesions first appear in early June as greenish-yellow spots that increase in size. They develop into characteristically brightly colored spots--the color can vary from yellowish-orange to red, depending upon the apple or crabapple cultivar (Figure 5 and 6). Symptoms are visible on both the upper and lower leaf surfaces (Figure 7).

Figure 5. Symptoms on red-pigmented crabapple (upper leaf surface).

Symptoms rarely develop on fruit and twigs. By mid-summer, minute "spore cups" called aecia develop at the edge of the lesions on the lower leaf surface.

Figure 6. Diagnostic symptoms of cedar-apple rust on leaves (upper leaf surface).

Figure 7. Symptoms on lower surfaces of crabapple leaves. Aecia are visible in these lesions.

The spores produced in these cups are called aeciospores. These spores are released from mid-summer into autumn. Aeciospores can only infect junipers and cannot infect other apples or crabapples. They are carried by wind and rain back to the juniper and red cedar hosts where they initiate another cycle of disease. When these spores land they germinate and stimulate the formation of galls. Galls are initially green to greenish-brown and gradually darken to brown as they age. It takes from 19-22 months to complete one life cycle of this fungus.

DISEASE MANAGEMENT:

Cedar-apple rust is not considered a life-threatening disease to either type of host so control measures are usually not necessary in most cases. However, if significant defoliation and/or fruit loss is experienced on apple or crabapple hosts, control measures may be necessary. This disease can be effectively managed through the combined use of culture, sanitation, resistance, and fungicide sprays. Cultural methods involve removal of either host within ½-1 mile from the other, although in most cases this is not feasible.

Sanitation involves pruning and removing galls from the red cedar and juniper hosts during the dormant season. Once again, this is practical in limited situations where only a few trees are involved and only a few galls are present.

Selection and planting of resistant cultivars or varieties is the most effective means of control since this effectively reduces or eliminates the occurrence of the disease. Examples of resistant apple cultivars include Delicious, Empire, Jonamac, McIntosh, and Paulared. Resistant crabapples include Ellwangerina, Henry Kohankie, Ormiston Roy, and Red Baron. Resistant junipers are: *Juniperus chinensis* var. *sargentii*, *J. communis* cv. Aureospica, and *J. virginiana* cv. Tripartita.

The final strategy for disease control involves the proper selection, timing, and application of fungicide sprays. Thorough coverage of all parts of the tree is necessary and the sprays should be applied until run-off. The fungicide label will contain

information on plant hosts and diseases, dosage rates, preharvest harvest interval (PHI), and safety precautions. Among the fungicides registered for use in Connecticut are chlorothalonil, mancozeb, triadimefon, propiconazole, and myclobutanil are registered for use and should be applied as necessary. An organic management option is sulfur. If harvesting fruit for consumption, please consult the fact sheet *Disease Control for Home Apple Orchards*. This guide contains information on fungicides registered for use on edible fruit.

Use of fungicides to protect *Juniperus* species has yielded disappointing results due to the difficulty in determining the timing of the applications since this mid-summer through fall infection period remains poorly understood. One of the only fungicides registered for use on junipers in the landscape is triadimefon.

May 2008 (revised)

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Diplodia Tip Blight: *Sphaeropsis sapinea*

Introduction

Dead, brown needles at the tips of pine branches may signal the presence of Diplodia tip blight. Surveys indicate that no species of pine (*Pinus*) is immune to this disease, although some species are more severely affected than others. Austrian pine (*Pinus nigra*) is one of the most severely damaged species in New York. Tip blight infection year after year can weaken and even kill large Austrian pine trees. Douglas-fir, white, Norway, and blue spruce may also be infected, but infection of these species usually develops only on trees that are injured or stressed and when infected pine are nearby to provide a source of inoculum (spores).

Figure 1: Symptoms of Diplodia Tip Blight (provided by **Dr. George W. Hudler, Cornell University)**

Symptoms and Signs

A few brown needles at the tip of the current season's growth are the first evidence of tip blight. These blighted needles are usually located on the lower branches of the tree. Needles that are in the early stages of development stop growing after infection and therefore appear stunted when compared to healthy needles. The number of blighted needles increases until the new growth for that year has been killed. This stunted and dead new growth on lower branches draws attention to the disease (**Fig. 1**).

Figure 2: Brown needles at the tip of the current year's growth. (provided by **Dr. George W. Hudler, Cornell University)**

With a hand lens you may see minute black fruiting bodies (pycnidia) at the base of diseased needles, especially under the papery leaf sheath. These are sure signs of this disease.

Symptoms of tip blight are not restricted to the needles. Future tree growth is often reduced by damage to or death of terminal buds during infection (**Fig. 2**). Twigs may be infected and become stunted or deformed. Stunted twigs may exude resin which traps blighted needles before they fall to the ground. Close examination of these twigs reveals cankers at

the first branch whorl and often near the leaf scars of blighted needles. Enlargement of these cankers results in the girdling of the twig and rapid death of the branch tip. In most cases, these symptoms are not observed beyond the current season's growth.

Disease Cycle

The fungus which causes tip blight of pine trees is *Sphaeropsis sapinea* (also known as *Diplodia pinea*). This fungus is present throughout the year in dead needles, leaf sheaths, twigs, and cones located either on an infected tree or on the ground. Small black fruiting bodies mature during late spring or early summer in this material. The brown oval spores ooze out of the fruiting structure during wet conditions and are scattered by wind, splashing rain, animals, or pruning equipment. Some spores land on young needles of the current season's growth. After the spore germinates, the fungus enters the needle through a stomate (or pore) and grows toward the base of the needle. A few hours later, a brown area develops near the point where the fungus entered the needle. By this time, the fungus has grown into the twig. The progressive invasion of the twig by the fungus results in browning of the attached needles and canker production in the twig.

Management Strategies

The damage caused by this disease is most severe on old or weakened trees. Old trees or trees exposed to unsuitable growing conditions, mechanical injury, or damage by insects may eventually be killed. Thorough weekly watering during extended dry periods of the growing season and spring broadcast fertilization will result in a tree that is more vigorous and more resistant to tip blight.

When only a few branches of a tree show symptoms of tip blight, a program of pruning and sanitation should help minimize disease. Prune off blighted twigs and destroy or discard them. Since fungal spores can be transported to healthy twigs during the pruning operation, prune when the tree is dry. Disinfect pruning tools before each cut by swabbing the cutting blades with a solution of 70% rubbing alcohol between each cut. Rake up all blighted needles, twigs, and especially cones which harbor the fungus and destroy or discard them.

Where disease is severe on smaller landscape trees, homeowners in New York State may apply some fungicides containing either potassium bicarbonate or propiconazole to try to minimize new infections. Follow label directions. Homeowners are discouraged from trying to apply pesticides to large landscape specimens, and should consider hiring a professional applicator if good cultural practices and fertilization do not sufficiently improve the overall health of such trees. Additional fungicides may be available for use by professional pesticide applicators.

Updated SLJ 2/15

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Dutch Elm Disease: *Ophiostoma novo-ulmi; O. ulmi*

Introduction

The rise of Dutch Elm Disease has been a devastating event in the history of tree diseases. It is caused by the fungi *Ophiostoma novo-ulmi* and *O. ulmi* and is vectored by bark beetles. The disease is referred to as "Dutch" Elm Disease because it was first described in Holland in 1921, although the pathogen originated in the orient. The disease began its destructive reign in the United States in 1930.

Prior to the arrival of this pathogen, many streets and parks were graced by the upright and spreading branch structure of American Elms. The appearance of a mature American Elm can make nature lovers stop and stare in awe. Due to the incredible interest in these trees, plant breeders have been trying to develop resistant American Elms for years. Unfortunately, many of those trees proved to be susceptible to other diseases such as Elm Yellows.

Figure 1: Characteristic streaking seen beneath the bark. (provided by Dr. Wayne Sinclair, Cornell University)

Symptoms and Signs

Symptoms develop quickly within a 4-5 week period and usually when the leaves have reached full size. The first visual symptom usually observed within the crown of the tree is referred to as "flagging". This occurs when one or more branches develop symptoms of wilting and/or yellowing of the leaves on a otherwise apparently healthy tree. Prior to this occurring, symptoms have developed internally and include the death of xylem cells, the loss of water conducting ability, and the browning of the infected sapwood in narrow streaks that follow the wood grain (**Fig. 1**). The fungus is present in the streaked wood, and isolations taken from this symptomatic tissue are needed to confirm infection by this pathogen. Occlusion of xylem vessels is due to the production of gums and tyloses. In the Western U.S. where summers are dry, water shortage and heat stress often mask symptoms.

Figure 2: Bark beetle galleries. (provided by Dr. Wayne Sinclair, Cornell University)

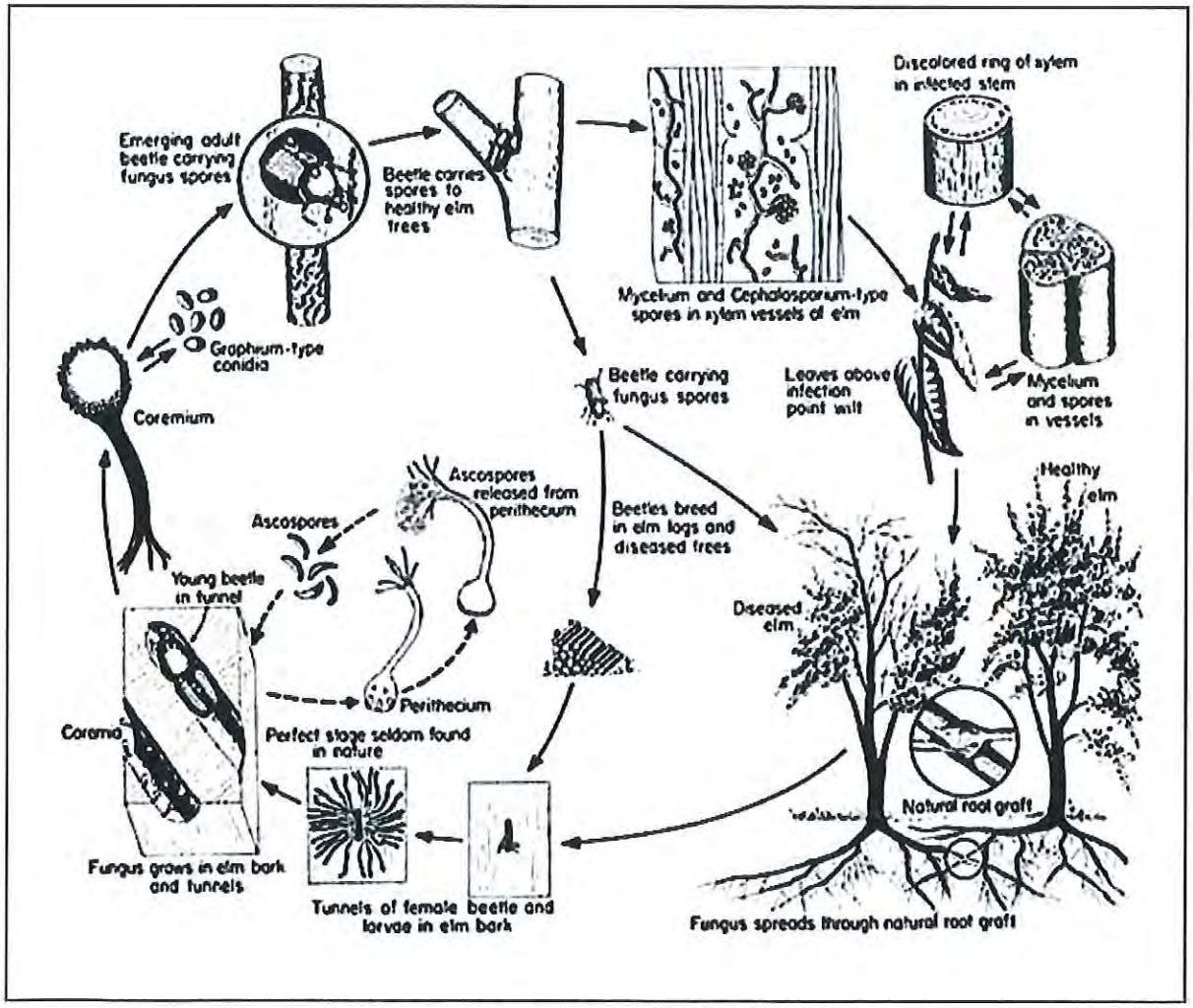
Disease Cycle

Spores of *O. novo-ulmi* are stored in xylem vessels and reproduce through budding. Dispersal of spores is via bark beetles that burrow under the bark and lay their eggs in wood galleries (**Fig. 2**). Elm bark beetles distribute *O. novo-ulmi* locally and over distances of several miles while the fungus may be distributed over longer distances in elm logs and in firewood. There are two species of beetle vectors known in North America, *Hylurgopinus rufipes* and *Scolytus multistratus.* Insects are attracted to healthy elms by volatile chemicals produced by the trees. Beetles bore into the inner bark and while feeding deposit spores of *O. novo-ulmi. Scotylus multistratus* feeds in the crotches of twigs, therefore, most infections occur in twigs. *Hylurgopinus rufipes* bores in the bark of branches and small trunks causing infections in major branches.

From the point of inoculation, the fungus moves upward and downward by two modes: in the liquid within xylem vessels and by the growth of fungal hyphae between xylem vessels after germination. *Ophiostoma novo-ulmi* reaches the roots within one season of infection where it continues to grow. The fungus grows in the roots and ascends the trunk in a wave of infection that kills the entire tree or a major part of it. Where elms are planted close together and there is a possibility of root grafting, *O. novo-ulmi* may move from one tree into the next through the roots. The fungus can also survive as a saprophyte in dead plant tissue.

Management Strategies

Management of DED requires a combination of strategies including sanitation, control of the insect vectors, removal of root grafts, preventative



Life cycle diagram of Dutch Elm Disease.

(provided by Dr. George Agrios from Plant Pathology, 3rd edition.)

fungicides, and use of resistant varieties.

1. Sanitation: Quick removal of diseased trees and symptomatic branches is necessary in managing this disease. Removal of the damaged parts reduces breeding sites for the elm bark beetle and removes the fungus from the area. Wood from infected trees can be used as firewood but should be used before the elm bark beetles emerge in the spring or should have the bark removed from the pieces prior to stacking. Branches with "flagging" symptoms should be removed making a cut well behind (5-10ft) any visual symptoms.

2. Insecticides to kill bark beetle vectors: Another option for management is to attempt to control the insect vector population by applying an insecticide. Timing of the application may depend on the type of insecticide you have selected. Some insecticides may target the spring feeding sites while others target the overwintering sites and should be applied during late autumn. Use of this management option should be considered carefully. It is difficult to attempt control over this beetle population due to the timing factor, the complete coverage issue, expense and pesticide exposure.

3. Root grafts: The fungus is capable of moving from tree to tree via root grafts. Trees planted within 25 to 50 feet of each other can easily develop grafted root systems. Breaking these root grafts is an important prevention measure but may be quite difficult for a homeowner to accomplish. A professional landscaper may have the necessary equipment, a vibratory plow, needed to perform this task. Breaking the root grafts prior to removing any diseased trees is recommended. Transmission of the pathogen may occur if the diseased tree's roots are pulled away or broken from the healthy tree's roots.

4. Preventative fungicide injections: Some fungicides are capable of protecting elm trees from infection, but this method of management should be considered very carefully. Fungicide injection must be performed by someone trained in the technique and may be expensive. Protection lasts only 1-3 years, and then must be repeated. In addition, some researchers are concerned that repeated wounding of the tree (by drilling holes) for the injection may open trees to decay.

5. Resistant cultivars: Developing or finding a truly resistant American Elm has been quite difficult. Originally the best method for creating a resistant American Elm was to cross it with closely related European or Asian species that are known to be resistant. Unfortunately, most of the resulting crosses do not carry the upright, spreading growth habit of the true American Elm and, therefore, are not well accepted as an alternative. Still, planting of resistant varieties derived from European or Asiatic elm species is advisable. In addition, after years of breeding and research, a number of clonal cultivars of American Elms with resistance have been made commercially available. These include 'Princeton', 'Independence', 'Valley Forge', 'New Harmony' and 'Jefferson'. Another group of resistant elms are collectively known as 'American Liberty Elm'. However, all of these elms are susceptible to Elm Yellows and should not be used where that disease occurs on native elm.

Created, KLS 1/01; Updated SLJ 1/15

READ THE LABEL BEFORE APPLYING ANY PESTICIDE! Changes in pesticide regulations occur constantly. All pesticides distributed, sold, and/or applied in New York State must be registered with the New York State Department of Environmental Conservation (DEC). Questions concerning the legality and/or registration status for pesticide use in New York State should be directed to the appropriate Cornell Cooperative Extension Specialist or your regional DEC office.

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Forest Health Care

Leaf Blotch of Horse-Chestnut

Leaf blotch of horse-chestnut is a leaf disease caused by the fungus *Guignardia aesculi.* It causes browning and curling of leaves of horse-chestnut and buckeye. The infection starts in early June and spreads throughout the summer in wet weather. The disease is not a serious health concern to trees.

Reddish brown blotches on the leaf.

Browning and curling of leaves.

Hosts and Damage

Host trees are horse-chestnut and buckeye.

The fungus may cause severe damage on young trees and nursery stock. It usually does not affect the health and growth of older trees except when they are stressed by drought, root loss or other factors. For infection and spread the fungus requires wet weather conditions.

Symptoms first appear as water soaked areas on young leaves early in summer. These lesions enlarge and may join together to form large reddish brown blotches on fully developed leaves later in summer. Tiny black spots (fruiting bodies) are present in dead areas. The infected leaves become dry and drop prematurely. The fungus overwinters on fallen leaves. In the spring, during wet weather, fungal spores are released from overwintered leaves to start a new infection on young unfolding leaves in the tree.

Specific Management Practices for Control of the Leaf Blotch of Horse-Chestnut:

- There is no effective control after the leaves are infected and the first symptoms appear. Prevention is the best method of control.
- Rake up and dispose of infected leaves in fall. This will reduce the source of infection the following spring. Leaves may be composted by City composting programs.
- Dense foliage preserves the moisture after rainfall creating favorable conditions for infection by the fungus. Crown thinning or selective pruning will improve air circulation and decrease humidity levels in a tree canopy.
- Avoid watering the leaves of the tree.

General Management Practices to Improve Plant Health:

- Water your trees during dry spells. Infrequent, but deep soaking preferably during the early morning hours is recommended. Water absorbing roots are located in the upper 25 cm of the soil and extend outward well beyond the canopy dripline.
- Place organic mulch, (e.g. wood chips), or living mulch, (e.g. ground covers) around the tree base to keep the soil moist for longer periods and encourage healthier roots.
- Avoid any unnecessary excavating, grade changes, soil compaction, root cutting or hard resurfacing around trees as these activities destroy vital roots which may lead to tree decline or death.
- Refrain from using high levels of salt or herbicides around trees.

Forest Health Care is a holistic approach to tree care that focuses on improving the health of trees in an urban environment. Our objective is a healthy, sustainable urban forest. Trees in urban forests are often stressed by compacted soil, drought, poor planting and pruning techniques, air pollution, road salt, damage from construction and much more. Trees planted in the right sites and properly maintained are less likely to suffer and are more resistant to pest problems.

Pest problems are managed using a decision making process that considers the following:

- Identification of the host and the pest.
- Monitoring of the host and the pest.
- Selection of the appropriate management strategy.
- Evaluation of the management plan.

Our focus is on pest management programs that are environmentally, socially and economically sound.

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Nectria Canker and Dieback

Red fruiting structures of nectria canker on beech (*Fagus*)

Worldwide, Nectria fungi cause several common canker and dieback diseases, especially in hardwood trees. Nectria canker, which is caused by the fungus, *Nectria galligena*, may occur on over 60 species of trees and shrubs including apple, ash, birch, dogwood, elm, sweet gum, holly, maple, pear and walnut. A similar disease infects members of the magnolia family. Nectria canker is usually not a fatal disease, but it can cause considerable damage as the cankered area is weakened and susceptible to breakage. It may also adversely affect the appearance of the affected plant. This disease is important commercially as it reduces the quality and quantity of forest products.

Another member of the *Nectria* genus, *Nectria cinnabarina*, causes the disease Nectria dieback. Also known as coral spot Nectria canker or Nectria canker, this disease occurs on many plant species, including apple, ash, barberry, birch, boxwood, crabapple, elm, hickory, honey locust, linden, maple, pear, rose and Japanese zelkova. *Nectria cinnabarina* usually grows as a saprophyte on dead wood, but if a plant is wounded or otherwise stressed, the fungus becomes an opportunistic weak parasite, producing cankers and causing dieback of twigs and branches. Maples are especially affected by this disease as are recently wounded or severely pruned trees and shrubs, urban ornamentals and new transplants of other species.

Symptoms and Diagnosis

Although it is most common in spring and fall, the Nectria fungus can infect plants throughout the year as long as there is sufficient moisture and the temperature is above freezing. Plants that are stressed by cold, drought, mechanical injuries or other disease are especially susceptible. Infections may be worse in autumn and winter when the host plant is dormant and wound recovery is weaker than in the growing season.

The first symptom of a Nectria infection is a depressed discolored area of bark near wounds or at the base of dead twigs or branches. These cankers are usually not noticed until other symptoms appear.

The first easily visible signs of Nectria canker are small creamy white or red to reddish orange fruiting structures and the development of callus tissue. This callus tissue is produced as the host plant attempts to isolate the fungus. If the callus does not isolate the infection, the fungus will continue to grow into healthy wood and the plant will respond by growing another ridge of callus tissue. This alternation of fungal growth and callus ridge, which may occur for many years, results in a rounded or elongated target-like shape. The bark of older ridges may decay and weather away exposing the ridges of wood underneath. This disease grows slowly and larger stems are rarely girdled, although multiple lesions may grow together and kill a branch or the entire plant. Plants that are stressed are most severely affected by the disease. This fungus may also affect apple fruit causing it to rot during storage.

Pests and Problems

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The first obvious sign of Nectria dieback may not occur until spring when the plant begins to grow. Affected twigs, branches, or even entire plants will not produce leaves or may wilt suddenly. Larger branches or small trees may be girdled and killed. The fungus produces reproductive structures that vary in color from creamy, coral pink, pink-orange, light purplish red or orange-red and that darken as they mature.

Life Cycle

Nectria galligena overwinters in the callus tissue growing slowly while its host is dormant. During moist periods, creamy white cushion-like fruiting structures will develop. These are followed by a second type of reproductive structure, which is red to reddish orange, pin-head sized and lemon-shaped, in autumn through spring. During rain or other moist weather, spores are released and dispersed by wind or water infecting susceptible plants through natural openings such as leaf scars or through wounds from improper pruning, sunscald, storm damage, frost cracks or other mechanical damage. As the fungus grows, it kills bark, cambium, and the outermost sapwood.

The life cycle of the Nectria dieback fungus is similar to that of Nectria canker. Creamy to coral pink to pink-orange or light purplish red spore-producing structures develop in spring or early summer. These will age to tan, brown, or nearly black. Orange-red fruiting structures, which mature to dark reddish brown and may persist through winter, are produced in summer and autumn. Both structures release spores that are dispersed by water and can invade susceptible tissue producing cankers and dieback.

Integrated Pest Management Strategies

1. Proper selection. Choose trees and shrubs that are well adapted to the climate of the area to minimize infection due to freeze damage and other environmental stresses.

2. Maintain plant vigor. Keep plants healthy and growing vigorously by using good cultural techniques. These include choosing the appropriate planting site, watering during dry periods, using mulch around the base of the tree or shrub and fertilizing and pruning properly. Pruning is best done in late winter. Avoid pruning in spring when higher moisture can increase risk of infection or in late summer and autumn, which can delay the plant's natural cold hardiness response. Minimize any wounding due to root pruning, transplanting or lawnmowers to reduce infection sites.

3. Prune. Prune out branch cankers during dry periods when conditions are unfavorable for infection. Disinfect pruning tools in a 1-part bleach to 9-parts water solution between each cut.

Organic Strategies

Strategies 1 and 3 are strictly organic approaches. Using an appropriate organic fertilizer would be a viable organic approach to Strategy 2.

More images:

Red-orange pustules of nectria canker on willow (*Salix*)

Red-orange pustules of nectria canker on willow (*Salix*)

Nectria canker on maple (*Acer*) trunk

Nectria canker on beech (*Fagus*)

Close-up of red fruiting structures of nectria canker on beech (*Fagus*)

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Rhizosphaera Needle Cast


[1]


[2]


[3]


[4]

Pest: Rhizosphaera Needle Cast (*Rhizosphaera kalkhoffii* and other spp.)

Rhizosphaera needle cast has become a major disease of landscape spruce (and sometimes fir) in recent years. The disease is caused by several species in genus *Rhizosphaera*, and while the ecology and pathogenicity of these species are poorly understood, the disease is most often attributed to *R. kalkhoffii*. Research from the UMass Plant Diagnostic Laboratory has identified at least nine phylogenetic species in the northeast from six host genera (*Picea*, *Abies*, *Tsuga*, *Pinus*, *Pseudotsuga* and *Cedrus*). Of these nine *Rhizosphaera* species, the majority of spruce are infected by one particular species. Infected trees are rarely, if ever, killed by the disease alone, but *Rhizosphaera* is an important contributor to decline when other stresses are present (e.g. drought, stem cankers, and insect pests). In addition, the premature needle shedding leads to decreased aesthetics and/or function of the trees as a privacy screen.

Host Plants:

Colorado blue spruce (*Picea pungens*), white spruce (*P. glauca*) and Oriental spruce (*P. orientalis*) are the most severely affected, while Norway spruce (*P. abies*) and red spruce (*P. rubens*) are more resistant to the disease. True fir (*Abies*), especially white fir (*A. concolor*), can suffer severe damage as well. Additional hosts in New England include pine (*Pinus*), hemlock (*Tsuga*), Douglas-fir (*Pseudotsuga*) and true cedar (*Cedrus*).

Symptoms & Disease Cycle:

Mild temperatures and prolonged needle wetness favor disease development. The pathogen invades susceptible needles through the stomata (pores used for gas exchange) and overwinters in diseased needles that have fallen to the ground or those that remain in the canopy. Beginning in the spring and lasting through the autumn season, spores are dispersed from infected needles by wind and splashing rainwater. Younger needles on interior portions of lower canopy branches are most commonly infected. In these locations air flow is limited, shade is more abundant and free moisture lingers on needles longer. Once needles are infected, symptoms may take 12 months or longer to become visible. When trees are stressed, symptoms of infection may develop more rapidly. On blue and white spruce, diseased needles often first appear purple, becoming brown to straw-colored. When the disease becomes well established in the lower canopy of a tree, *Rhizosphaera* then spreads upward in successive years, gradually leading to increased rates of premature needle shedding. In certain settings, mature trees can be severely defoliated, leaving only a small tuft of live foliage in the upper canopy. Pycnidia (small, black-colored fruiting bodies of the fungus through which spores are discharged) develop on the surface of infected needles and can be observed with the naked eye or a 10X hand lens during almost any season. They appear as fuzzy black, sooty dust. On spruce, the pycnidia are present on all sides of the needle, whereas on fir, they occur only on the underside of the needle.

Management:

Conifers vary in their susceptibility to the disease, so if disease pressure in the landscape is high, choose resistant trees such as Norway spruce for new plantings. The disease is more destructive on spruces planted in shaded settings or in tight hedgerows. Most spruces are shade intolerant and prefer full sun with no surrounding plants to thrive. Environmental stresses such as drought, deep planting and mechanical root injury contribute to disease severity. Additionally, opportunistic stem cankering fungi and insect pests also contribute to disease severity by reducing host vigor. When branches in the lower canopy die, they should be pruned out to limit the establishment and spread of opportunistic pathogens. Fungicides can be effective in certain cases but will likely have little impact once the fungus is well established in the canopy. Chemicals labeled for use in the landscape against *Rhizosphaera* include: azoxystrobin, chlorothalonil, chlorothalonil + thiophanate-methyl, copper salts of fatty and/or rosin acids, copper hydroxide, copper hydroxide + mancozeb, mancozeb, thiophanate-methyl and Phosphorous acid. Fungicides should be applied in the spring when new needles are half-elongated and then again once on regular intervals should wet weather. It should be kept in mind that infections can take place anytime during the growing season when environmental conditions allow.

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Topics: Commercial Horticulture
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VERTICILLIUM WILT OF ORNAMENTAL TREES AND SHRUBS

Verticillium wilt is a common disease of a wide variety of ornamental trees and shrubs throughout the United States and Connecticut. Maple, smoke-tree, elm, redbud, viburnum, and lilac are among the more important hosts of this disease. Japanese maples appear to be particularly susceptible and often collapse shortly after the disease is detected. Plants weakened by root damage from drought, waterlogged soils, de-icing salts, and other environmental stresses are thought to be more prone to infection.

Verticillium wilt is caused by two closely related soilborne fungi, *Verticillium dahliae* and *V. albo-atrum*. Isolates of these fungi vary in host range, pathogenicity, and virulence. *Verticillium* species are found worldwide in cultivated soils. The most common species associated with Verticillium wilt of woody ornamentals in Connecticut is *V. dahliae*.

SYMPTOMS AND DISEASE DEVELOPMENT:

Symptoms of Verticillium wilt vary by host and can be characterized as acute or chronic. Plants or branches with acute infections may wilt and die suddenly (Figure 1).

Figure 1. Japanese maple with acute symptoms of Verticillium wilt.

They also develop a variety of symptoms that include wilting, curling, browning, and drying of leaves. These leaves usually do not drop from the plant. In other cases, leaves develop a scorched appearance, show early fall coloration, and drop prematurely (Figure 2).

Plants with acute infections start with symptoms on individual branches or in one portion of the canopy. This symptom is often called "flagging." Symptoms are often located on one side of the plant, which can be diagnostic for this vascular wilt disease (Figure 3). As the fungus grows and spreads within the plant from year to year,

symptoms appear in more of the canopy. Symptoms are usually most obvious in mid to late summer or fall--frequently after stressful periods of hot, dry weather. In some cases, infected plants suddenly die the season following initial symptoms.

Figure 2. Verticillium-infected Japanese maple leaves with marginal scorch symptoms.

Figure 3. Maple with a portion of the canopy exhibiting symptoms of premature fall coloration.

Plants with chronic infections usually show general decline, as exhibited by sparse canopies consisting of undersized, off-colored leaves, poor growth and vigor, and branch dieback. Infected plants can occasionally produce heavy crops of seeds or samaras. Plant death can be slow or sudden, depending upon the extent of infection and general plant health.

Another diagnostic characteristic of Verticillium wilt is distinctive discoloration or streaking in the sapwood. The color of the discoloration varies by host. For example, it is dark olive-green in maple (Figure 4), chocolate brown in redbud, and brown in elm. Streaking can be random in infected wood--it is not reliably present in small-diameter branches or twigs. However, discoloration is more consistent near the bases of larger, symptomatic branches. Since many other fungi can cause similar discolorations of sapwood, positive diagnosis of Verticillium wilt requires culturing sapwood tissue in the laboratory.

Figure 4. Diagnostic vascular discoloration in sapwood of maple infected with Verticillium wilt.

Verticillium dahliae is soilborne and persists for indefinite periods of time in the soil as resting structures called microsclerotia. Germination and growth of these structures is stimulated by exudates from a host plant

or from decaying organic matter. The fungus enters the roots and the water transport system (xylem) of the plant. It then grows, sporulates, and moves systemically throughout the plant. Spores (conidia) are ovoid and are borne on specialized hyphae (phialides) in a whorl around a conidiophore. Verticillium is named for this verticillate or whorled arrangement of the phialides on the conidiophore (Figure 5). The presence of the fungus in the xylem restricts movement of water and nutrients by its physical presence as well as through production of enzymes and toxins. As the fungus grows in the xylem, the plant responds in both physical and biochemical ways to contain or compartmentalize the fungus. This results in plugging and gumming of water-conducting vessels, which further restricts water in the host.

Figure 5. Verticillate spore-bearing structures on conidiophores.

This ongoing interaction between the host and the fungus can help to explain reports of symptom "remission" in some infected plants. Symptoms appear to stop and start for a year or so, although in most cases they reappear and encompass more of the plant canopy. This can be understood as an external expression of what is happening inside the infected plant--symptoms disappear after the plant has successfully compartmentalized the fungus to keep it from spreading. Symptoms reappear when the fungus breaks through the compartmentalization barriers and begins to grow and move again in the host.

Verticillium wilt occurs on a wide range of woody and herbaceous hosts. However, to date, all gymnosperms and monocots appear to be resistant or immune to this disease. A fact sheet "Verticillium Wilt of Vegetables and Herbaceous Ornamentals" has more information on the effects of the disease on nonwoody hosts.

MANAGEMENT STRATEGIES:

Managing Verticillium wilt is most successful using a multifaceted strategy. There are no satisfactory controls for this disease once plants are infected.

- Fungicides are not effective for control.
- For plants that exhibit mild symptoms, it can help to maintain vigor by following sound cultural practices. These include pruning symptomatic branches, watering during periods of drought, and fertilizing (based on a soil test). Mulching is also helpful since it helps maintain soil moisture, moderate soil temperatures, and minimize chances for mechanical injuries. Although infected plants cannot be cured, these practices can sometimes delay the progression of disease for several years.
- *Verticillium* fungi can survive for many years as microsclerotia in the soil. Therefore, it is necessary to avoid planting susceptible species in the areas known to be infested. In such cases, gymnosperms and resistant or immune species should be planted (Table 1).
- Since microsclerotia can be present in soil or debris, it is important to avoid moving soil or debris from areas of known infection.
- As a precaution against spread, all tools should be disinfested between cuts with

a 10% solution of household bleach, 70% alcohol, or one of the commercial products such as Physan 20.

- Several studies have demonstrated that microsclerotia can be found in fresh wood chips from infected trees. Although the studies are inconclusive, it is probably best to avoid using fresh chips to mulch susceptible hosts. It is suggested that chips be composted for at least one year prior to use.

Table 1. Resistance of Selected Woody Ornamentals to Verticillium Wilt

Resistant or Immune	Susceptible
Apple *(Malus)*	Ash *(Fraxinus)*
Arborvitae *(Thuja)*	Azalea *(Rhododendron)*
Beech *(Fagus)*	Barberry *(Berberis)*
Birch *(Betula)*	Black Locust *(Robinia)*
Boxwood *(Buxus)*	Box Elder *(Acer negundo)*
Butternut *(Juglans)*	Boxwood *(Buxus)*
Crabapple *(Malus)*	Catalpa *(Catalpa)*
Dogwood *(Cornus)**	Cherry, other stone fruits *(Prunus)*
Fir *(Abies)*	Coffee tree, Kentucky *(Gymnocladus)*
Firethorn *(Pyracantha)*	Currant *(Ribes)*
Gingko *(Gingko)*	Dogwood *(Cornus)**
Hackberry *(Celtis)*	Elm *(Ulmus)*
Hawthorn *(Crataegus)*	Honeysuckle *(Lonicera)*
Hickory *(Carya)*	Lilac *(Syringa)*
Holly *(Ilex)*	Linden *(Tilia)**
Honey Locust *(Gleditsia)*	Magnolia *(Magnolia)*
Hornbeam *(Carpinus)*	Maple *(Acer)*
Juniper *(Juniperus)*	Redbud *(Cercis)*
Katsura tree *(Cercidiphyllum)*	Rose *(Rosa)*
Larch *(Larix)*	Russian Olive *(Elaeagnus)*
Linden *(Tilia)**	Serviceberry *(Amelanchier)**
Mountain Ash *(Sorbus)*	Smoke tree *(Cotinus)*
Mulberry *(Morus)*	Spirea *(Spirea)*
Oak *(Quercus)*	Sumac *(Rhus)*
Pear *(Pyrus)*	Viburnum *(Viburnum)*
Pine *(Pinus)*	Weigela *(Weigela)*
Poplar *(Populus)*	Yellowwood *(Cladratis)*
Serviceberry *(Amelanchier)**	
Spruce *(Picea)*	
Sweet Gum *(Liquidambar)*	
Sycamore *(Platanus)*	
Walnut *(Juglans)*	
Willow *(Salix)*	
Yew *(Taxus)*	

* The resistance or susceptibility of these plants will depend upon the cultivar of the tree and the strain of *Verticillium* present in the soil.

May 2008

White Pine Blister Rust

Cronartium ribicola
J. C. Fisch

Elliptical cankers covered with yellow blisters that release infectious spores to be carried away by the wind. *Courtesy of Tracey Olson, PDA*

Hosts

- Eastern white pine (*Pinus strobus*)
- Other five-needled pines

Alternate Hosts

- Currants
- Gooseberry
- Other *Ribes* species

Damage Potential

- Moderate–high

Symptoms and Signs

On Pine

Year-round:

- Chlorotic needles, stunted growth
- Chlorotic, dead, or dying tree tops or branches
- Yellow-bordered cankers on trunk or 3- to 4-year-old branches; oozing resin or rodent feeding may be apparent
- Resin flow on main trunk that hardens to white, orange, or brown

Early spring:

- Orange-yellow blisters breaking through cankered bark to release spores

Late spring to early summer:

- Sticky, yellow fluid produced from yellow-brown blister on canker; fluid will blacken after a short time

Fall:

- Patches of yellow or brown bark on young growth; area may be swollen but progresses to spindle-shaped canker by second year of infection

On *Ribes* Species

Summer:

- Orange spores on underside of leaf; upper surface may have yellow, diffused spots

Late summer to early fall:

- Brown, hairlike projections on underside of leaf

Causes of Similar Symptoms

- Fungal root rots
- Pales weevil
- Pine root collar weevil

Identification

White pine blister rust is the only stem rust of five-needled pines. It requires an alternate host (*Ribes* species) for new infections to occur on pine.

Early detection can be difficult due to very subtle symptoms during the first year of infection. First, a small, yellow or red spot appears on the needle at the site of infection and eventually the needle may die as the fungus grows into the bark tissue. The newly infected bark tissue will become brown with a yellow border surrounding a section of dead needles. The stem may be slightly swollen.

During the second year of infection and beyond, long, elliptical-shaped cankers develop on branches and the main trunk as the fungus advances. Cankers can eventually girdle the branch, which results in "flagging" (Fig. 1), or they can girdle the trunk, killing all growth above the canker. Yellowish-green bark tissue may be visible around the canker. Blisters (0.25 inch, 3 mm across), spores, and resin flow all arise from the canker area. Rodent feeding may be evident around this area as well.

Biology and Life Cycle

The life cycle of white pine blister rust may take 3–6 years to complete. It begins in late summer or early autumn when basidiospores from the alternate host (*Ribes* species) are wind and rain dispersed, entering the pine needle through the stomata (Fig. 2). Basidiospores may be carried in wind currents for up to a mile.

Figure 1. Flagging as a result of cankers girdling a branch. ***Courtesy of John W. Schwandt, USDA Forest Service, Bugwood.org (#1241718)***

Figure 2. Yellow/red infection site on a needle. ***Courtesy of USDA Forest Service Ogden Archive, Bugwood.org (#1467424)***

Figure 3. Elliptical cankers formed after the first year of infection. *Courtesy of Joseph O'Brien, USDA Forest Service, Bugwood.org (#5061066)*

Figure 4. Mass of powdery, yellow spores covering the surface of the canker (spring). *Courtesy of Tracey Olson, PDA*

Figure 5. Sticky, yellow liquid exuded by a canker just before hardening off. *Courtesy of Joseph O'Brien, USDA Forest Service, Bugwood.org (#5042098)*

The fungus grows into the bark tissue at a rate of 5–6 inches (12.5–15.0 cm) per year and begins to form cankers after the first year of infection (Fig. 3). In spring, 3–4 years after the initial infection, pale yellow or cream-colored blisters (aecia) rupture through the bark of active cankers (Fig. 4). They release powdery, yellow spores (aeciospores) that are carried in the wind over long distances to the alternate host and cause infection. The aeciospores can only infect *Ribes* species. After the spores are released, the cankered area on the pine remains swollen and roughened. In summer, the sticky, yellow fluid that exudes from the site hardens and leaves small, brown-rust-colored scars (Fig. 5).

On the alternate host, the aeciospores enter the stomata of the leaf during wet weather. Diffused, yellow spots become visible on the upper leaf surface soon after infection occurs. Within a few weeks, pustules form on the leaf underside and release spores that repeatedly infect the same plant or other *Ribes* in the vicinity (Fig. 6). This repeating stage serves to increase the levels of inoculum. In late summer, small, brown, hairlike structures appear on the underside of the leaf. Eventually, basidiospores are produced and wind dispersed back to susceptible pines in the vicinity.

Figure 6. Underside of *Ribes* leaf with fruiting bodies. *Courtesy of Robert L. Anderson, USDA Forest Service, Bugwood.org (#0355052)*

Monitoring and Management Strategies

Plantation Establishment

- Northeastern Pennsylvania is especially prone to white pine blister rust.
- Do not plant white pine in low-lying areas where cool, moist air is likely to settle.
- Do not plant white pine species if there is an abundance of alternate hosts (*Ribes* spp.) in the surrounding area.
- Consider removing alternate host material from within 1,000 feet of white pine plantings.

Preseason

- Tag trees suspected to have white pine blister rust to check for blisters in mid-April.

Growing Season

- Scout trees for blisters in mid-April.
  - Randomly select at least 50 trees that are 5–10 years of age.
  - Look for yellow or orange blisters on branches and trunks.
  - Also scout tagged trees.
- If 10 percent or more of sampled trees are infected:
  - Prune/remove cankers from trees before infection reaches trunk.
  - Control alternate host before August.
- Remove and destroy trees with trunk infections.
- Inspect trees throughout the year for cankers.
- At the end of the season, evaluate results and update records.

Control Options

Biological

- No recommendations are available at this time.

Mechanical

- Prune and destroy all branches with cankers.
- Remove and destroy all alternate host plants in and around plantation within a minimum of 1,000 feet.

Biorational

- No recommendations are available at this time.

Chemical

- Apply an appropriate fungicide in late summer to protect pines from infection from basidiospores released by an alternate host.
- Apply an appropriate herbicide to control the alternate host.

Next Crop/Prevention

- Inspect plants/nursery stock; buy from a reputable company.

Arborist Study Guide
Plant Pest List

Insects

1. Birch Leafminer
2. Bronze Birch Borer
3. Browntail Moth
4. Cooley Spruce Adelgid
5. Eastern Tent Caterpiller
6. Elongate Hemlock Scale
7. Fall Webworm
8. Gall Mites on Deciduous Trees
9. Gypsy Moth
10. Hemlock Woolly Adelgid
11. Japanese Beetle
12. Mountain Ash Sawfly
13. Pine Bark Adelgid
14. White Pine Weevil
15. Winter Moth

Diseases

1. Anthracnose
2. Black Knot of Plum and Cherry
3. Cedar-Apple Rust
4. Diplodia Tip Blight
5. Dutch Elm Disease
6. Leaf Blotch of Horse Chestnut
7. Nectria Canker
8. Rhizosphaera Needlecast
9. Verticillium Wilt
10. White Pine Blister Rust

Environmental Problems

1. Drought and Winter Injury
2. Herbicide Damage

Abiotic Diseases Of Woody Ornamentals

Biotic diseases involve fungi, bacteria, viruses, nematodes, etc., and abiotic diseases involve non-living things. Of the two major types of disease, abiotic diseases are by far the most important ones on landscape and nursery plants.

Abiotic diseases are the result of the interaction, over an extended period of time, between the plant and one or more factors such as lack of space for root growth, the presence of chronic or acute levels of air or water pollutants, or the presence of extremes of moisture, heat, light, soil pH, and nutrients. Most abiotic diseases cause generalized symptoms to develop over a large portion of the tree or shrub. Wilting, yellowing, the development of smaller than normal leaves, slowing of growth, branch death, water sprout (epicormic shoot) formation, premature autumn leaf coloration, and heavier than normal seed production are some of the symptoms characteristic of abiotic diseases. Sometimes very similar symptoms develop in a location on very different species of plants. This is another indication that abiotic factors rather than living pathogens are involved in damaging plant health. Below is a brief review of abiotic diseases other than those caused by pollution.

Moisture Extremes

Plants require sufficient moisture to grow but not so much that the roots drown. Related to this is the fact that soil compaction by heavy equipment or foot traffic in the plant's root zone reduces the pore space in the soil. Moisture holding capacity and pore space for air in the soil are reduced. Also, percolation of moisture through the soil profile is disrupted so that the desirable air/moisture balance is not maintained.

Roots die from a lack of oxygen. To diagnose problems caused by moisture extremes, refer to local weather records to determine if rainfall has been lacking or excessive. Determine if the plants are being irrigated, how the water is applied and how frequently it is applied. Use a soil probe or shovel to determine if the root zone and soil profile are wet or dry. Check the drainage pattern in the vicinity. Runoff may be being directed toward the plant. Or excavation may have changed the pattern in a way that now prevents moisture that had once been available to the plant to now be directed away from the site. If a plant has been recently transplanted or if nearby excavation has greatly pruned away roots, drought symptoms can develop even if the surrounding soil is moist. There may not be enough roots present to support the above-ground portion of the plant. Drought is one of the most important abiotic stresses on plants in the landscape and nursery.

Nutrient Extremes or Imbalances and pH

Plants need nitrogen, phosphorus, potassium, magnesium and many other major and minor nutrients in sufficient quantities to grow but not in excess such that one or more becomes toxic to the plant. There must also be a balance among the available nutrients. When there is an imbalance among certain elements (such as calcium/magnesium) the plant selectively takes up one to the exclusion of other element, thereby causing a deficient or toxic level in the plant. Symptoms can vary by species of plant affected, the time when the deficiency or toxicity began, and other growing condition factors. The soil pH plays a very important role in determining whether a given element is tied up in the soil or is available to the plant. If the soil pH is either too high or too low, some elements will be unavailable while others may be available in toxic quantities. Positive diagnosis of nutrient problems must be based on the analysis of representative leaf and soil samples.

Heat/Light Extremes

The effects of light and heat are difficult to separate. Plants in locations with very high light intensity are exposed to high temperature conditions. Both high light intensity and high temperature can result in the breakdown of chlorophyll in the leaves.

Leaves on the sunny side of the plant or those receiving the high temperature may exhibit death of the tissue at their margins or in large areas between the veins. High light intensity and temperature can scald and kill the cambium, the growing cell layer just under the bark, of thin-barked trees, trees newly transplanted or trees freshly pruned. The wood shrinks as it dies and dries. The bark cracks open and predisposes the damaged area to attack by canker causing fungi. Eventually, a branch dieback develops. Freezing is one of the most important abiotic factors in the landscape. Uneven light absorption by tree trunks in the winter results in uneven thawing of water in the plant. If the temperature in the trunk drops rapidly, the quick freezing and expansion of the water

splits the bark of the tree opening it to attack by wood rotting and canker-causing fungi. Even if bark splitting does not occur, the damage to the wood may result in branch death, delayed bud break, and lack of flowering. In extreme cases, the plant may be killed to the soil line. Late spring frosts often kill young, tender foliage and flowers. This is diagnosed by correlating local weather records to the onset of symptoms. Succulent tissue may turn brown or black after freezing. Often, a large numbers of the outer twigs are affected. However, the death of the tissue occurs over a short period and does not continue if frost is the cause. Usually, vegetative buds farther back on the twig develop.

Inhibition of Root Development

Plants need space for their roots to grow. As roots mature, their cell walls thicken and become suberized and lignified to provide structural support and probably to resist attack by insects, fungi, bacteria, nematodes, and other organisms in the soil. The young portions of the roots are responsible for most of the uptake of water and nutrients. If the tree does not produce new root tips because root growth is restricted, plant growth slows. In some cases, this is beneficial because the plant will take much longer to outgrow its location. In the interior landscape or outdoor patios for example, it is not desirable to have a plant grow rapidly because it will overwhelm its setting, require increasing amounts of water and nutrients, and need to be repotted frequently or replaced by a smaller plant. Trees that normally attain a large size may be placed under a great deal of moisture stress if their roots are restricted. This "drought" stress predisposes them to root rots, cankers, and branch diebacks. Containerized plants can be checked for root circling in the container. Similarly in plants in restricted locations such as parking lot islands, a soil probe or shovel can be used to determine where the roots are located. An examination of the weeds or lack of plant growth around a plant can indicate how much foot traffic occurs. Some weeds grow primarily where foot traffic damages its less-resistant competitors. The force needed to insert a soil probe or metal rod into the root zone soil can be compared to the force required to penetrate the soil in an area away from the root zone as an indication of soil compaction.

General Diagnosis

These are a few examples of abiotic diseases. Some requirement of the plant is not met and the result is a decline in plant health. Determining exactly which requirement is not being met demands that all the symptoms be noted and that the plant site be carefully examined. Rule out the possibility that any organisms present on the plant are the primary cause of the disease. Sending a sample to a plant disease clinic is necessary in order to obtain a positive identification of the organisms found. It is known that certain organisms ONLY cause disease when the plant is under some type of stress or when the plant has been damage first by some other factor. In general, canker-causing fungi most readily damage trees that are under drought stress. The stress must be relieved. Keep in mind that, as noted above, drought may be caused by lack of moisture or damage to roots that results in lack of uptake of available moisture. This example shows the complexity of abiotic disease diagnosis.

Because ABIOTIC diseases are the ones that occur most frequently on plants in the landscape, it is critical that landscapers, nurserymen, and grounds maintenance personnel develop the skills and experience needed to diagnose them.

Epicormic shoots (water sprouts)

Branch dieback

Early fall leaf color

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Code: XL0001

Pest Alert

DROUGHT AND WINTER DRYING

FOREST SERVICE--U.S. DEPARTMENT OF AGRICULTURE
IN COOPERATION WITH YOUR STATE AGENCY

Drought and winter drying have periodically caused major damage to trees. Drought reduces the amount of water available in the soil. In the case of winter drying, the water may be in the soil, but freezing of the soil makes the water unavailable to the tree. In both cases, more water is lost through transpiration than is available to the plant.

Symptoms of drought and winter drying may include partial or complete leaf scorch, premature leaf drop, or death of the entire tree. De-icing compounds can cause similar damage. If the tree is not killed, it may be stressed enough to make it more susceptible to attack by fungi such as *Armillaria mellea and Diplodia pinea,* or by bark beetles, Ips spp.

Tip burn

Marginal and interveinal burning

Drought-caused mortality

Partial mortality caused by winter drying

Winter drying

Reports of injury due to drought should be sent to

* U.S. GOVERNMENT PRINTING OFFICE: 1977 - 751-726

BARTLETT TREE EXPERTS
SCIENTIFIC TREE CARE SINCE 1907

Diagnosing and Preventing Herbicide Injury to Trees

Bruce R. Fraedrich, Ph. D., Plant Pathologist

Herbicides have become an integral part of landscape maintenance because chemical weed control often is more economical and efficient than hand or mechanical cultivation. Chemical weed control also is a primary method of vegetation management in industrial areas, utility rights-of-way, along highways and other non-cropland areas. This increased dependence on herbicides in landscape maintenance has caused concern and confusion among arborists regarding the effects of herbicides on non-target woody plants. Much of this confusion stems from the multitude of herbicides, formulations and products presently on the market. Herbicide injury also is difficult to diagnose based on symptoms alone because other environmental or cultural adversities, as well as infectious diseases and insects, can produce similar symptoms. Detection of herbicide residues by chemical analysis is expensive and often unfeasible.

This report presents a listing of herbicides frequently used in the landscape and their potential toxicity (or lack thereof) to woody plants. A description of herbicide injury symptoms is presented along with suggestions to aid diagnosis and recommendations for remedial treatment of herbicide-injured plants.

TYPES OF HERBICIDES

Pre-Emergence Herbicides For Turf - This group of herbicides is widely used in lawns to prevent growth of annual grasses, such as crabgrasses and certain annual broadleaf weeds. The principal herbicides included in this group are: **DCPA (Dacthal), benslide (Betasan), siduron (Tupesan) and benefin (Balan)**. These herbicides also are mixed with fertilizers to form the so-called "weed and feed" products for turf. Since pre-emergence herbicides for turf have little or no root activity, they are not phytotoxic to woody plants when properly applied.

Pre-Emergence Herbicides For Landscape Plantings - The following herbicides are registered for pre-emergent control of annual weeds around woody ornamentals in the landscape and in commercial nurseries:

Chloramben (Ornamental Weedar)
oxyzalin (Surflan)
dichlobenil (Casoron)
pronamide (Kerb)
diphenamide (Enide)
simazine (Prenceep) (low rates)
napropamide (Devrionol)
trifluralin (Treflan)
oxadizon (Ronstar)

The herbicides generally are not absorbed by roots of woody plants and are safe around most species. Exceptions exist and labels of specific

pre-emergence herbicides should be consulted concerning precautions. Drift contacting the foliage may cause injury on some ornamental species especially if leaves are wet. Some pre-emergence herbicides may cause injury on tolerant woody ornamentals if mis-applied at exceptionally high rates. Injury is usually evident as foliage chlorosis followed by leaf browning and defoliation. Plants usually recover from injury from these herbicides.

Post-Emergence Grass Killers - This group of herbicides is composed of arsonates such as **MSMA and DSMA** which are applied to turf for post-emergent control of crabgrass and other annual grasses. Arsonates are not root-absorbed and are safe around woody vegetation. Drift contacting the foliage of desirable woody plants may cause leaf burn, but injury is confined only to treated foliage. Plants usually refoliate and recover from injury caused by drift from arsenate herbicides.

Post-Emergence Broadleaf Herbicides - The following herbicides selectively kill broadleaf plants but are safe on most grass species: **2, 4-D 2, 4-DP (dichlorprop), MCPP (mecoprop), dicamba (Banvel) and picloram (Tordon). 2,4-D, 2,4-DP and MCPP** commonly are referred to as growth regulator or hormonal type herbicides because their chemistry resembles naturally occurring growth regulators in plants. The hormone herbicides are the most widely used chemicals for broadleaf weed control in turf and are the principal herbicides that are mixed with turf fertilizers to form the "weed and feed" products.

At rates recommended for broadleaf weed control in turf, the hormone herbicides are not root active and their persistence in soil is less than one month. Subsequently, **2,4-D, 2,4-DP, and MCPP** seldom will injure woody plants when applied at their labeled rates for weed control in turf. Granular formulations are safest near trees because there is less likelihood of drift than from spray formulations.

Drift from these herbicides, which contacts the foliage of desirable plants, will cause leaf distortion and browning; however, most woody plants will tolerate the injury and recover. On warm days certain forms (ester formulations) of **2,4-D** may volatilize and cause minor leaf injury when applied near desirable woody plants. Using amine formulations of **2,4-D** minimizes the possibility of injury from volatilization. At high rates, the hormone herbicides are absorbed through the roots of woody plants, which can cause severe injury. Shallow-rooted species and deciduous hardwoods are most sensitive to high rates of the hormone herbicides while conifers are more resistant.

Dicamba and picloram have a different chemistry than the hormone herbicides, but they produce similar injury symptoms. Dicamba is used for controlling hard-to-kill broadleaf weeds in turf and to control woody vegetation in rights-of-way, woodlots, fence lines, and other non-cropland areas. Dicamba is absorbed through the roots of woody plants and can severely injure or kill ornamentals if applied within their root zone. This herbicide is persistent in soil for three months or more and will leach downward in most soil types. The persistence and mobility of dicamba increases the likelihood of the herbicide contacting absorption roots of trees. Injury on woody plants can range from leaf distortion and defoliation to branch dieback and complete death, depending on the rate and frequency of application and the species, age and vigor of the plant.

Picloram (Tordon) is applied primarily for broadleaf weed and brush control in non-cropland such as utility rights-of-way. Picloram is absorbed through the foliage and roots of plants and will severely injure or kill trees if applied within their root zone. At rates applied for brush control, picloram may have a residual effectiveness of a year or more in most soils.

Post-Emergence, Non-Selective (foliar absorbed) Herbicides - This group of herbicides which includes **paraquat, glyphosphate (Round-Up), aminotriazole (Amitrole), and cacodylic acid** controls both broadleaf weeds and grasses. These herbicides are absorbed through the foliage of plants and have little or no root activity. These materials are safe around woody plants as long as drift does not contact the foliage, green bark or suckers of desirable species.

Non-Selective Soil Sterilant Herbicides - Soil sterilants are used for total vegetation control in rights-of-way, pipelines, industrial facilities and other non-cropland areas.

Herbicides included in this group are:

linuron (Lorox)
hexazinone (Velpar)
bromacil (Hyvar)
diuron (Karmex)
tebuthiuron (Spike)
karbutilate (Tandex)
atrazine (Aatrex) (high rates)
prometon (Pramitol)
monuron (Urox)
princep (Simazine) (high rates)

The soil sterilant herbicides are extremely toxic to woody plants and misapplications of this group account for the majority of herbicide-related deaths of landscape plants. Soil sterilants are absorbed readily by tree roots and translocated throughout the crown. Many of these herbicides are water-soluble and may be leached or carried by runoff into the root system of a desirable plant. These materials also have a long residual of a year or more. Subsequently, plants may be injured long after application if roots grow into a treated area or if a tree or shrub is planted in contaminated soil.

Injury and mortality of landscape plants is particularly common when soil sterilants are used for total vegetation control for sidewalks and driveways around homesites before pavement is installed. Soil sterilants often are formulated with other herbicides and sold under a variety of trade names. Homeowners and inexperienced applicators often do not read the labels or comprehend the acute phytotoxicity of these materials.

SYMPTOMS OF INJURY

Symptoms of herbicide injury vary considerably depending primarily on the material applied and the dosage rate. Herbicide injury cannot be diagnosed solely on symptomatology because other adversities can produce a similar effect.

Hormonal (Growth Regulator) Herbicides - **(2,4-D, 2,4-DP, MCPP, dicamba, picloram.)** Symptoms are most pronounced on new growth. Leaf distortion including cupping, curling, abnormal elongation of leaf margins (epinasty) and parallel leaf venation are key symptoms. Current year's shoots may be twisted and flattened rather than round or angular. Leaf necrosis, dieback and mortality may be evident in severe instances especially with dicamba and picloram. Late spring frosts may cause leaf and twig distortions similar to injury by growth-regulator herbicides.

Triazine Herbicides ***(simazine, prometon, atrazine)*** - The initial symptom is chlorosis of leaf blade between the veins. Leaf browning, dieback and death may follow. Nutrient deficiencies, air pollution injury, and sucking insects may produce leaf chlorosis similar to triazine herbicide injury.

Contact Herbicides ***(glyphosphate, paraquat, cacodylic acid)*** - Light drift may appear as small, brown spots where droplets contacted the foliage. Leaf browning and dieback usually is restricted to treated areas of the plants. Foliage, such as root suckers, accidentally treated late in the growing season may cause severe crown-dieback the following year, especially on shrubs and small trees.

Amino-traizole causes severe chlorosis and may bleach treated foliage white. Amitrole may be translocated to untreated portions of the plant where similar symptoms are evident. In severe instances, dieback and decline may result.

Urea and Uracil Herbicides (Soil Sterilants: ***bromacil, tebuthiuron, diuron, monuron, karbutilate, linuron)*** - Initial symptoms are chlorosis of leaf blades and/or veins. Necrosis of the leaves progresses from the margin. Branch dieback and mortality usually follows. Other factors, which can produce leaf browning, and dieback similar to injury from soil sterilants include natural gas, lightning strikes, deicing salts, winter drying, transplanting shock and infectious diseases.

DIAGNOSIS

Before making a diagnosis, the arborist must have full knowledge of the appearance of the healthy plant. Many ornamental plant varieties are selected for yellow or white variegation in the leaves, which could be mistaken for herbicide injury if the arborist is unfamiliar with the plant. Some varieties also are selected for contorted foliage and stems such as the fantail and corkscrew willow, contorted white pine and contorted European filbert.
These varieties could be mistaken for hormone-type herbicide injury by the unsuspecting arborist.

Diagnosis of herbicide injury requires careful observations and detailed questioning of property owners. If herbicide injury is suspected, check the weeds in the lawn and other near-by vegetation for similar symptoms. Herbicides usually produce the same symptoms on a wide range of plant species, which is unusual for other causal agents. If injury symptoms are widely present, check for the source of the herbicide such as the lawn, rights-of-way, industrial area, fence line or highway. Question the property owner or applicator as to the type of herbicide applied, the rate and application method. Often the herbicide container must be inspected because the applicator is unfamiliar with the material or the exact rate of application. Also remember that herbicide injury may result from applying insecticides or fungicides to desirable plants using an herbicide-contaminated sprayer.

If a root-active herbicide such as **dicamba** or soil sterilant material was applied, consider the fact that tree roots typically extend far beyond the dripline. Recent research has shown that absorption roots may extend more than two times the height of the tree from the trunk. Tree roots may absorb an herbicide even if the material was applied outside the dripline of the plant. Soil sterilant herbicides also have long residual activity and most are water-soluble. Subsequently, these materials may leach into the root zone of a desirable plant long after they were applied. Phytotoxicity also may occur when planting in herbicide-contaminated soil.

REMEDIAL TREATMENT OF HERBICIDE INJURED TREES

Trees usually recover from light herbicide injury. Irrigating the plant during dry periods will minimize moisture stress, which may hinder recovery.

Irrigation also will help leach root-active herbicides from the root zone of the plant. Fertilization should be avoided for a minimum of one growing season following injury, because stimulating excess growth can compound injury from certain herbicides. Similarly, if branch dieback results, pruning should be delayed for a least a year to fully assess the extent of the injury. This will avoid additional pruning of dead branches that may result from continued decline. However, immediate pruning is necessary if dead branches pose a danger to life or property.

If root active chemicals such as **dicamba**, high rates of **2,4-D** or **soil sterilants** are applied near trees, activated charcoal may help tie-up herbicide residues and minimize injury. Activated charcoal must be applied immediately following the mis-application of the herbicide, preferably before the onset of symptoms. If tree roots absorb the herbicide and damage becomes evident, activated charcoal will be of little value in ensuring survival of the affected plant.

Activated charcoal is applied at 150 times the amount of the active ingredient per acre of the applied herbicide. For example, if four pounds active ingredient of an herbicide were applied per acre, then 4 X 150 or 600 pounds of activated charcoal per acre would be necessary to deactivate the residue. For best results, charcoal should be soil incorporated by tilling it into the upper three to four inches of soil, or by

mixing with water and injecting it as slurry using a high volume sprayer.

Trees, which are seriously declining from herbicides generally, do not recover and removal usually is required. Trees should be left standing for at least one growing season after the damage has occurred to fully assess the potential for recovery. Replanting the site should be attempted only after herbicide residues have degraded.