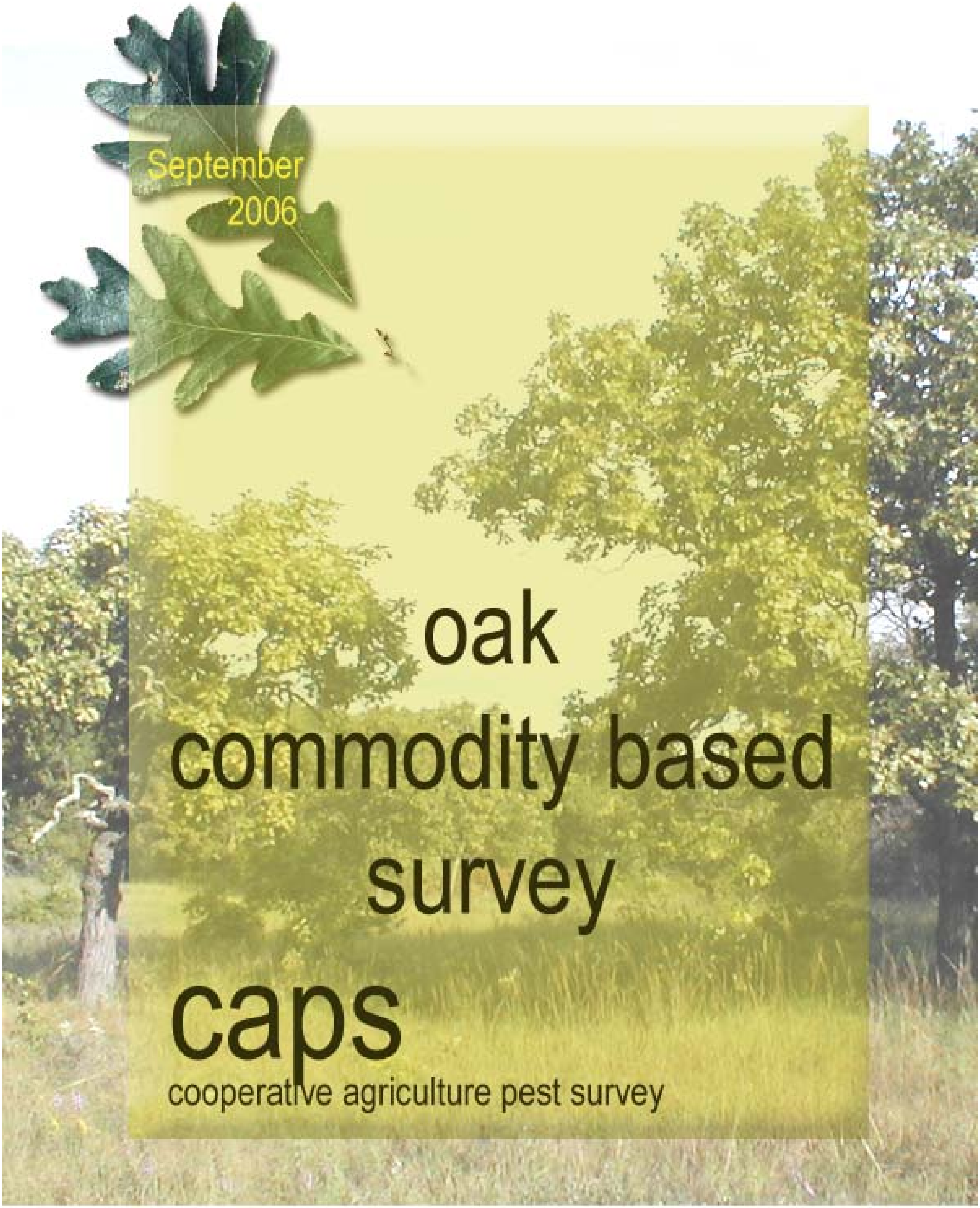

September
2006

oak
commodity based survey

caps
cooperative agriculture pest survey

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On the cover: An oak savannah forest at Cedar Creek Natural History Area: A National Science Foundation Long Term Ecological Research Site,
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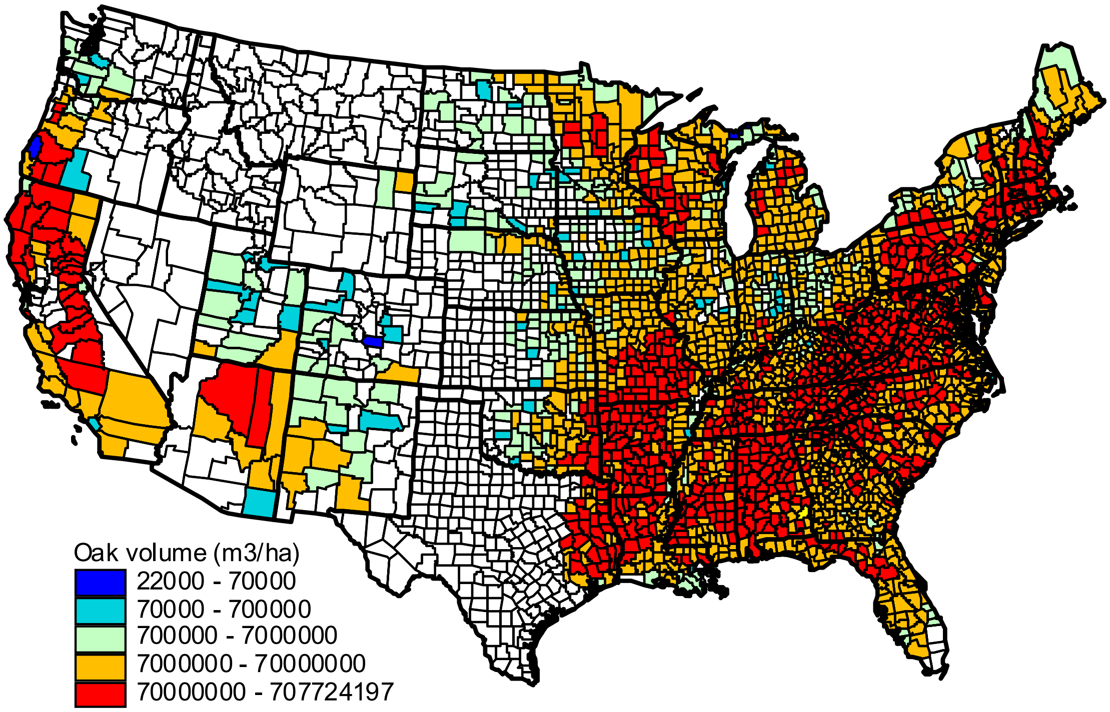
Introduction



Oaks (*Quercus* spp.) are one of the most diverse groups of native plants in the US. At least 85 species occur in the country. In the western US, *Quercus* is one of four prevalent genera of hardwoods, which together account for 17% of 238 million acres of forest land; in the eastern U.S., oak forests account for 52% of 384 million acres of forest land. Oak trees and forests are valued in urban and natural areas for wildlife habitat, recreation, and for their beneficial effects on soil, air and water quality. Declining health and increasing mortality of oaks are already major concerns in several areas of the country. Seventeen *Quercus* species are identified as threatened or endangered.

In the US, hardwood species provided 36% of the round wood products harvested in 2001. Oaks contributed more than 117 billion cubic feet (14%) of the total growing stock on timberland in 2002 and 95% was from the eastern US. Species in the white oak group make up 43% of the growing stock volume in the eastern US. From National Forests alone, almost 17 million board feet of oak, worth over $50 million, were sold in 1997. Sixteen percent came from white oaks. In 1997, the US exported 1.2 billion board feet of hardwood lumber, 70% of which went to countries on other continents.

The US imports a substantial volume of oak logs and lumber annually. In 2005, 16.5 million cubic meters of red and white oak logs and 14.5 million cubic meters of raw (not dried) red and white oak lumber were imported. In 2005, the volume of imported red oak logs was at a five year high, up 82.5% from 2004.

The majority of these imports were from Canada. Nevertheless, trade in raw oak may accidentally introduce new insects and pathogens to the United States or contribute to the spread of non-native species that currently have a restricted distribution on the continent.

This document addresses 20 non-native species that have the potential to adversely affect oaks. Most of these species do not occur in the United States. However, a few do. Established non-native species continue to present risks to oak forests as they spread into previously unaffected areas. Additionally, new geographic variants of established species may arise and impact oaks in ways not previously known. All pests were considered a serious threat by the Cooperative Agricultural Pest Survey or the North American Forestry Commission.

In this document, we present biological details that are relevant to the survey and detection of each species. The following information can be found within each subsection:

Pest Description - a description of the morphology of the species. In some cases these descriptions are quite technical, but are intended to help provide reliable identifications. For technical accuracy, most descriptions are quoted directly from taxonomic authorities.

Biology and Ecology - a summary of the life history of each species with a particular emphasis on when various life stages may be present and active.

Pest Importance - a description of the ecological and economic impact each species may have. Impacts are not limited to oak. In some cases, pests are particularly problematic in agriculture. This illustrates the ability of agricultural pests to thrive in forests and vice versa.

Symptoms - a description of damage the species may cause on a host plant. This may be useful for surveying oak stands.

Known Hosts - a complete listing of plants which have been reported to support the growth and reproduction of the species.

Known Distribution - a list of countries from where the species has been reported.

Potential Distribution within the US - a summary of regions within the US that may have suitable environmental conditions for the species.

Survey - available techniques that have been used to detect the species. This section also describes other species that might be confused with the target pest.

Arthropods
Beetles & Weevils

Aeolesthes sarta

Scientific Name

Aeolesthes sarta Solsky

Synonyms:
Pachydissus sartus Solsky
Aealesthes sarta Gahan
Aeolesthes sarta Gahan
Aelesthes sarta Stebbing
Aeolesthes sarta Beeson & Bhatia
Aeolesthes sarta Beeson

Common Names

City longhorned beetle
Town longhorned beetle
Sart longhorn beetle
Uzbek longhorned beetle
Quetta longhorned beetle

Type of Pest

beetle, wood borer

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda
Order: Coleoptera, **Family:** Cerambycidae

Reason for inclusion in manual

Exotic Forest Pest Information System – classified as a very high risk pest with the potential to attack oaks

Fig. 1 Adult male *Aeolesthes sarta*.
[Image from M. Hoskovec, http://www.uochb.cas.cz/~natur/cerambyx/aeolsarta.htm]

Pest Description

"Adult beetle large elongate, cylindrical, steel grey in colour, elytra covered with thick coating of pubescence, shining white when newly emerged disappearing with age. [Note that USDA (1968) and Sengupta and Sengupta (1981) describe the color as reddish brown.] Body length vary [*sic*] from 22 to 42.2 mm in males and 29.2 to 43 mm in females. Antennae more than double the body length in males and shorter than body in females. Elytra obliquely truncate at the apex, the outer angle being unarmed and the sutural angle dentate or shortly spined" (Ahmad et al. 1977).

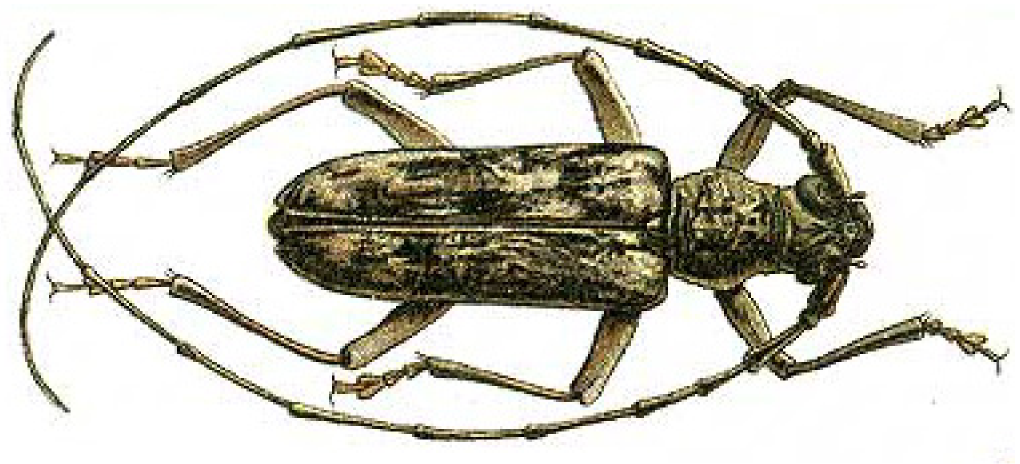


Fig. 2. Drawing of *Aeolesthes sarta* adult male
[Image from www.zin.ru/animalia/Coleoptera/rus/jacobs65.htm]

"Larvae – Length about 6 mm in the first stage but nearly 80 mm when fully grown. Yellowish and about 15 mm when full grown" (reviewed in USDA 1968).

Biology and Ecology

Little is known about the biology of *A. sarta*, primarily because this pest is often concealed inside a host plant (Duffy 1968, reviewed in USDA 1968, Ahmad et al. 1977). Much of the biology of *A. sarta* was described by Ahmad et al. (1977) from a laboratory study using poplar logs.

Aeolesthes sarta completes its life cycle in two years (Ahmad et al. 1977). Adults emerge in April to early May when the average daily temperature reaches 20°C [68°F] (reviewed in EPPO 2005). Beetles are active at night until early morning and seek shelter during the day in old excavated tunnels, pupal chambers or under bark (reviewed in EPPO 2005). Beetles are infrequent fliers, and adults typically remain on the surface of the host on which they developed. Mating takes place between 8 hrs to 2 days following emergence. Males can mate multiple times.

Oviposition begins 1 to 5 days following emergence and lasts about 1-3 weeks. Females make slits in the bark of the trunk or large branches and deposit eggs in groups of 1-13. One female produces up to 270 eggs (Ahmad et al. 1977), but 50 eggs is more typical (Duffy 1968). Viable eggs are produced at a minimum temperature of 15°C [59°F], however oviposition does not occur at temperatures of 10°C [50°F] or above 35°C [95°F] (Ahmad et al. 1977). Maximum egg hatch occurs at 22-24°C [72-75°F], 12-13 days following oviposition. Adult males live 7-15 days and females live 19-25 days (Ahmad et al. 1977).

Larvae hatch within 10-17 days (Ahmad et al. 1977). Early stars (up to 1 month old) form galleries just beneath the bark. Later instars penetrate into the sapwood. Towards the end of the first season of development, larvae bore up a tree about 25 cm [~10 in] along the long axis of the trunk or branch and then turn downward to form a gallery approximately 15 cm [~6 in] long (reviewed in EPPO 2005). Larval galleries will be filled with wood dust and frass. The larva overwinters at the base of the downward gallery protected by a thick plug constructed from wood borings (reviewed in EPPO 2005).

In the following spring, larvae continue to feed, making progressively deeper tunnels. At the end of July, larvae prepare pupation cells that are once again protected by a thick plug constructed from wood borings. The pupal stage lasts approximately 4 months. The adults stay in the pupation cells for 1-2 months overwinter then emerge in the spring through a round exit hole.

Pest Importance

Aeolesthes sarta is a polyphagous, stem-boring longhorn beetle and an economically important pest of forest, ornamental and fruit trees throughout its range (Duffy 1968, reviewed in USDA 1968, Sengupta and Sengupta 1981, reviewed in Orlinski 2000, reviewed in EPPO 2005). Ahmad et al. (1977) call *A. sarta* “one of the most destructive borer[s] of poplar.” Similarly, Gaffar and Bhat (1991) list this beetle as “one of the most destructive pests of nut trees” in India. *Aeolesthes sarta* is also a pest of economic concern in Kashmir and Iran (Duffy 1968, Farashiani et al. 2001). *A. sarta* is known to attack stressed and apparently healthy trees, and as few as 1-3 larvae per tree can cause mortality (reviewed in Orlinski 2000). In Quetta, Pakistan a severe infestation of *A. sarta* in 1904-06 severely injured 5000 trees (Duffy 1968).

Larvae make extensive galleries in the cambium and sapwood, and this boring activity will often kill a tree (Ahmad et al. 1977). Even if the infested tree is not killed outright, the wood is of limited commercial value due to the galleries and borer holes (Ahmad et al. 1977, Gaffar and Bhat 1991). Larval tunnels cause the tree to dry out and the branches to break off readily in wind (reviewed in USDA 1968). An infestation of *A. sarta* typically causes tree death in 2-4 years (reviewed in USDA 1968, Thakur 1999).

Risks associated with *A. sarta* for North American forests have been evaluated previously. Orlinski (2000) considered the insect to pose a very high risk, but this assessment was very uncertain. The potential for establishment, spread, economic injury, and environmental damage were each rated high, but these evaluations assume that North American hardwood species would be suitable.

Symptoms

Occasionally, larvae of *A sarta* eat so much of the cambial tissues that the overlying bark falls off the tree (Ahmad et al. 1977, reviewed in Orlinski 2000). Extensive feeding in the cambium can lead to girdling, branch die back and subsequent death of the tree (Duffy 1968, USDA 1968, Ahmad et al. 1977, reviewed in Orlinski 2000). Evidence of infestation also includes wood borings near the surface of the entry hole or at the base of the host tree, and round exit holes (Ahmad et al. 1977, reviewed in EPPO 2005).

Known Hosts

Aeolesthes sarta attacks a wide range of deciduous tree species:

Hosts	**References**
Acer sp. (maple)	(Gressitt 1951, Duffy 1968, Orlinski 2000, EPPO 2005)
Acer cultratum (bakimu)	(Ahmad et al. 1977, Sengupta and Sengupta 1981)
Aesculus sp. (buckeye)	(Gressitt 1951)
Aesculus indica (Indian horse-chestnut)	(Duffy 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981)
Alnus subcordata (Caucasian alder)	(Farashiani et al. 2001)
Betula sp. (birch)	(Orlinski 2000, EPPO 2005)
Carya sp. (pecan)	(Gaffar and Bhat 1991)
Castanea sp. (chestnut)	(Gaffar and Bhat 1991)
Corylus colurna (Turkish hazelnut)	(Gressitt 1951, Duffy 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981)
Cydonia sp. (quince)	(Duffy 1968, USDA 1968, Gaffar and Bhat 1991)
Elaeagnus sp. (elaeagnus)	(Orlinski 2000, Farashiani et al. 2001, EPPO 2005)
Fraxinus sp. (ash)	(Duffy 1968, Orlinski 2000, EPPO 2005)
Gleditsia sp. (locust)	(Orlinski 2000, EPPO 2005)
Juglans sp. (walnut)	(Gressitt 1951, USDA 1968, Gaffar and Bhat 1991, Orlinski 2000, EPPO 2005)
Juglans regia (English walnut)[1]	(Duffy 1968, Yagdyev and Tashlieva 1976, Ahmad et al. 1977, Sengupta and Sengupta 1981, Orlinski 2000, Farashiani et al. 2001, EPPO 2005, Mir and Wani 2005)
Malus sp. (apple)	(USDA 1968, Gaffar and Bhat 1991, Orlinski 2000, EPPO 2005)
Malus pumila (=*M. domestica*) (paradise apple)[1]	(Ahmad et al. 1977, Orlinski 2000, EPPO 2005)
Malus sylvestris (=*Pyrus malus*) (European crabapple)	(Duffy 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981)
Morus sp. (mulberry)	(Duffy 1968, Orlinski 2000, Farashiani et al. 2001, EPPO 2005)
Platanus sp. (plane)	(Gressitt 1951, Ahmad et al. 1977, Orlinski 2000, EPPO 2005)

Hosts	**References**
Platanus acerifolia (=*P. hybrida*, =*Platanus ×hispanica*) (London planetree)	(Orlinski 2000, EPPO 2005)
Platanus orientalis (Oriental planetree)	(Duffy 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981, Orlinski 2000, Farashiani et al. 2001, EPPO 2005)
Populus sp. (poplar)	(Gressitt 1951, USDA 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981, Thakur 1999, Orlinski 2000, EPPO 2005)
Populus alba (white poplar)	(Duffy 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981, Orlinski 2000, Farashiani et al. 2001, EPPO 2005)
Populus ciliata (Himalayan poplar)	(Ahmad et al. 1977)
Populus diversifolia (huyang)	(Orlinski 2000, EPPO 2005)
Populus euphratica (Euphrates poplar)	(Duffy 1968, Yagdyev 1975, Ahmad et al. 1977, Orlinski 2000, EPPO 2005)
Populus nigra (Lombardy poplar)	(Duffy 1968, Ahmad et al. 1977, Farashiani et al. 2001)
Populus talassica (Talas poplar)	(Orlinski 2000, EPPO 2005)
Populus × canadensis (*P. deltoides × nigra*) (=*P. × euramericana*) (Carolina poplar)	(Ahmad et al. 1977, Orlinski 2000, EPPO 2005)
Prunus sp. (stone fruit)	(Gressitt 1951, Duffy 1968, Gaffar and Bhat 1991, Orlinski 2000, EPPO 2005)
Prunus amygdalus (=*Amygdalus communis*) (almond)	(Mustafa and Janjua 1942, Duffy 1968, USDA 1968, Ahmad et al. 1977, Gaffar and Bhat 1991, Farashiani et al. 2001)
Prunus armeniaca (apricot)	(Duffy 1968, USDA 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981, Gaffar and Bhat 1991)
Prunus racemosa (bird cherry)	(Duffy 1968, Ahmad et al. 1977)
Pyrus sp. (pear)	(Gressitt 1951, Ahmad et al. 1977, Gaffar and Bhat 1991, Orlinski 2000, EPPO 2005)
Pyrus communis (pear)	(Duffy 1968, Ahmad et al. 1977)
Quercus sp. (oak)	(Orlinski 2000, EPPO 2005)
Robinia sp. (locust)	(Duffy 1968, Orlinski 2000, Farashiani et al. 2001, EPPO 2005)
Salix sp. (willow)	(Gressitt 1951, USDA 1968, Ahmad et al. 1977, Sengupta and Sengupta

Hosts	**References**
	1981, Orlinski 2000, Farashiani et al. 2001, EPPO 2005)
Salix acmophylla	(Duffy 1968, Orlinski 2000, EPPO 2005)
Salix alba (white willow)	(Duffy 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981)
Salix babylonica (weeping willow)	(Duffy 1968, Ahmad et al. 1977)
Salix songarica	(Orlinski 2000, EPPO 2005)
Salix tetrasperma	(Duffy 1968)
Salix turanica	(Orlinski 2000, EPPO 2005)
Ulmus sp. (elm)	(Gressitt 1951, Duffy 1968, USDA 1968, Ahmad et al. 1977, Orlinski 2000, Farashiani et al. 2001, EPPO 2005)
Ulmus minor (European field elm)	(Orlinski 2000, EPPO 2005)
Ulmus pulmila (dwarf elm)	(Orlinski 2000, EPPO 2005)
Ulmus wallichiana (Himalayan elm)	(Duffy 1968, Ahmad et al. 1977, Sengupta and Sengupta 1981)

1. Listed in Orlinski (2000) as a “preferred host”.

Known Distribution

Aeolesthes sarta occurs in the Palearctic and Orient (Duffy 1968). It has specifically been reported from:

Location	**References**
Afghanistan	(Duffy 1968, USDA 1968, Ahmad et al. 1977, Orlinski 2000, EPPO 2005)
China	(Gressitt 1951, USDA 1968)
India	(Duffy 1968, USDA 1968, Sengupta and Sengupta 1981, Gaffar and Bhat 1991, Thakur 1999, Orlinski 2000, EPPO 2005, Mir and Wani 2005)
Iran	(USDA 1968, Ahmad et al. 1977, Orlinski 2000, Farashiani et al. 2001, EPPO 2005)
Japan	(Orlinski 2000)
Kazakhstan	(Duffy 1968, USDA 1968)
Kyrgyzstan	(Duffy 1968, Orlinski 2000, EPPO 2005)
Malaysia	(Orlinski 2000)
Pakistan	(Mustafa and Janjua 1942, Gressitt 1951, Duffy 1968, USDA 1968, Ahmad et al. 1977, Orlinski 2000, EPPO 2005)
Sri Lanka	(Orlinski 2000)

Location	References
Tajikistan	(Duffy 1968, Orlinski 2000, EPPO 2005)
Turkmenistan	(Duffy 1968, Yagdyev and Tashlieva 1976, Orlinski 2000, EPPO 2005)
Uzbekistan	(Duffy 1968, USDA 1968, Orlinski 2000, EPPO 2005)

Potential Distribution within the US

The potential distribution of this insect in the US is difficult to predict. Based on the worldwide distribution of the species, *A. sarta* seems to be more closely associated with biomes classified as montane grassland and tropical and subtropical moist broadleaf forest. Montane grassland does not occur in the US and subtropical moist broadleaf forest only occurs in southern Florida. The insect may occur in temperate broadleaf and mixed forests, but the information from Asia is not detailed enough to know for certain.

In general, the species is most problematic in areas that are hot and dry (reviewed in CAB 2006).

Survey

Visual inspection of trees is the only practical method to survey for this insect; no attractants have been identified. Exit holes in tree trunks or large branches are one of the most obvious symptoms (EPPO 2005). Dust from larval boring may be present at the base of a tree (Orlinski 2000). Trees may have large areas with apparently rotting bark (Mustafa and Janjua 1942). Infested trees may also show symptoms of dieback (EPPO 2005).

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Agrilus biguttatus[*]

Scientific Name

Agrilus biguttatus Fabricius

Synonyms:
Buprestis biguttatus (=*biguttata*), Fabricius, 1777
Agrilus pannonicus Piller & Mitterpacher, 1783
Agrilus subfasciatus Ménétriés, 1832
Agrilus morosus Gory & Laporte, 1837

A complete list of proposed or unavailable synonyms is also documented by Jendek (2002) and Silfverberg (1977).

Common Names

Oak splendour beetle
Two-spotted wood borer

Type of Pest

Wood borer, phloem feeder

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Coleoptera, **Family:** Buprestidae

Reason for inclusion in manual

CAPS Priority Pest (FY 2007)

Pest Description

Fig. 1. Adult *Agrilus biguttatus*: (typical length is 8-13 mm; 5/16-1/2 in.)
[Image from Gyorgy Csoka, www.invasive.org]

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[*] This document is largely excerpted from the report: Davis, E.E., S. French, and R.C. Venette. 2005. Mini-Risk Assessment: Metallic Beetle, *Agrilus biguttatus* Fabricius [Coleoptera: Buprestidae]. Available on-line at www.aphis.usda.gov/ppq/ep/pestdetection/pra/ abiguttatuspra.pdf

Female. “Length 10 ½ mm; breadth (across the shoulders of the elytra) 2 ½ mm ... The front of the head flattened and with a central depression. ... The elytra long and narrow, at the base slightly broader than the thorax, and the sides sharply convergent from the middle to the apices; flattened over the disc, convex along the sides, and with two incurved basal hollows between the prominent long shoulders and the flattened scutellar area; the elytral surface more shagreened than rugulose in appearance, and with two small white-haired spots adsutural and towards the apex. Abdomen with six white-haired spots ventrally. Coloration bright metallic bronze green with localised suffusion of violet. The under-surface finely and more or less closely punctate with considerable confluence of the punctures on the thoracic parts” (Staig 1940).

“The short head is brilliant bronze green and rugulosepunctate. ... The prominent oval eyes are vertically placed and wide apart; their finely faceted corneal surfaces are dull green with darker patches and some small golden spots. The short antennae are metallic dark bronze green and are serrate, except the first three segments and the last or eleventh which are club-shaped; the first or basal segment is the largest, the second and third are about equal in size” (Staig 1940).

“The pronotum is transverse, its breadth (2 ¼ mm) is greater than its length, which measures 1 ½ mm; it is broadest across the middle and the base is slightly narrower than the front. ... The sides of the pronotum (viewed from above) are rounded and have arcuate narrow rims (the lateral carinae); as the carinae are obliquely placed, the sides are considerably deflected in front and the sharp anterior angles are low down at the gena and at a short distance from the lower ends of the eyes. ... The general appearance of the surface is that of a shagreened sculpture with slightly rugulose effect. The coloration of the pronotum is metallic bronze green suffused with violet” (Staig 1940).

“The prosternum, dark bronze green, is roughly triangular; its base is emarginate and sinuous and is marked off from the middle portion, by a deep transverse furrow, as a distinct gular part. ... The metasternum is dark bronze green and irregularly punctate, with very short and fine whitish hairs; but most of the punctures are confluent in broken lines. The surface is convex, except over the ante-coxal area, where it is flattened and depressed” (Staig 1940).

“The scutellum is bright metallic bronze green tinged with violet; it is large and triangular, but the sides are rounded at the base and are deeply incurved towards the apex and upon the large transverse base there is a strong median transverse ridge or carina” (Staig 1940).

“The elytra are bright metallic bronze green with violet along the narrowly deflected sides and upon the apices. Between the middle and the apices, and close together at the sutural margins, there are two small and irregular white spots, these being slight depressions with overlying silvery white recumbent hairs of considerable length. The length of the elytra (8 ½ mm) is more than three times the breadth (2 ½ mm across the shoulders), which is slightly greater than that of the pronotum across the middle. ... The surface is punctate and more shagreened than rugulose in appearance" (Staig 1940).

"The short legs are uniformly dark metallic bronze green, brighter on the tibiae, and the leg surface is finely punctulate with very short and fine whitish hairs. ... The abdomen is uniformly dark metallic bronze green with a strong suffusion of violet. The proximal sternum (first and second sterna conjoined) is very long, very nearly half the length of the abdomen. ... There are six white spots (slight depressions of the surface covered with moderately long overlying silvery-white hairs) on the third, fourth and fifth sterna, one pair on each and antero-lateral in position" (Staig 1940).

Male. "...length is 11 ½ mm. The anterior tibiae have a small sharp hook at the distal end on the inner side. The suffusion of violet on the elytra is more extensive than in the female metatype" (Staig 1940).

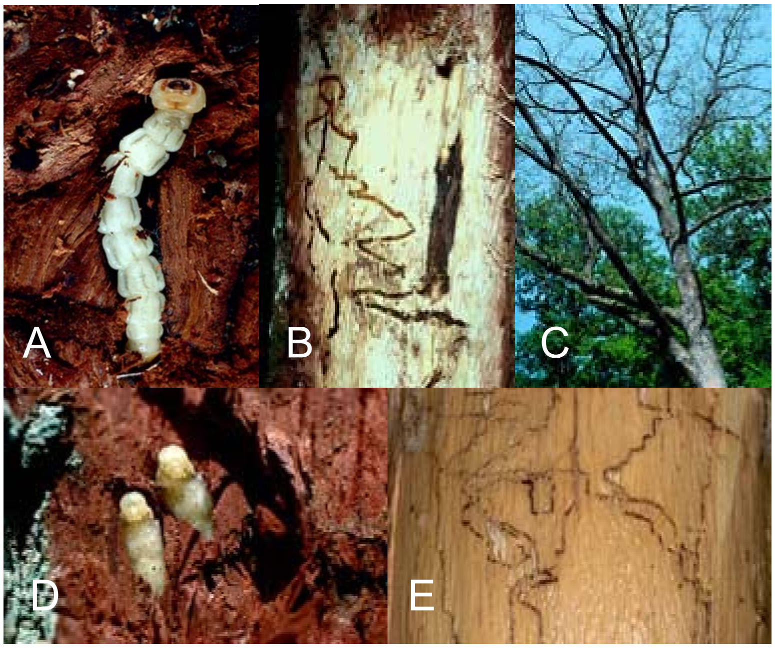

Fig. 2. Life stages and damage caused by *Agrilus biguttatus* (A) larva tunneling in *Quercus robur*; (B) cambium necrosis with characteristic "zig-zag" gallery pattern; (C) tree death resulting from *A. biguttatus* infestation; (D) pupae visible in bark; and (E) "stair" gallery pattern. Images are not to scale.
[Images from Louis-Michel Nageleisen, www.invasive.org].

Biology and Ecology

Agrilus biguttatus has 1 to 2 generations annually. Complete development typically occurs over a two-year period (Ciesla 2003, Vansteenkiste et al. 2004). *Agrilus biguttatus* preferentially oviposits in fresh, moist host material with plenty of sun exposure, particularly within the thinning crown of a declining host. Older, larger diameter trees, 30-40 cm [12-16 inch] diameter at breast height (dbh), are also preferred (Hackett 1995a, Moraal and Hilszczanski 2000b, Vansteenkiste et al. 2004). Successful colonization of a host tree may depend on larval population density because numerous borers can overcome tree defenses (Vansteenkiste et al. 2004).

Adults emerge from D-shaped holes (2.5-4 x 2-3 mm) between May and August (Key 1991, Hackett 1995a, Moraal and Hilszczanski 2000b, Vansteenkiste et al. 2004). Peak emergence occurs in June and July. Shortly after emergence, adults feed on foliage in the tree crown before taking flight. Females lay clusters of 5 or 6 eggs in bark crevasses from May to early August. In the field, eggs typically hatch in 1-2 weeks (Vansteenkiste et al. 2004). Larvae bore longitudinally through the bark, then proceed toward the inner bark, cambial layer, and outer sapwood, making a zig-zag patterned gallery (Fig. 2). Feeding occurs in the cambial layer of trees or in the stems of small woody plants (Ciesla 2003, Vansteenkiste et al. 2004). Larvae overwinter inside the bark for 1-2 winters, followed by pupation (Vansteenkiste et al. 2004). *Agrilus biguttatus* completes 5 larval instars.

Pupation occurs in the spring (April-May) of the second or third year. Pupae develop in individual cells that are 10.4-14.4 mm long and 3.0-4.5 mm wide. (Hackett 1995a, Moraal and Hilszczanski 2000b). Newly eclosed adults will remain in the bark for two weeks before emerging.

Pest Importance

Agrilus biguttatus is a significant pest of oak forests in eastern and western Europe, Russian Asia, northern Africa, and the Middle East (Jacquiot 1976, Gutowski and Lugowoj 2000, Moraal and Hilszczanski 2000b, 2000a, Hilszczanski and Kolk 2001, Vansteenkiste et al. 2004). Over 20,000 ha (nearly 50,000 acres) of oak mortality has been attributed to this insect in the Voronej region of Russia alone. The insect also caused considerable damage in several regions of France between 1945-1949 following notably hot and dry summers (Jacquiot 1976).

In Europe, *A. biguttatus* is an environmental concern (Key 1991). In England, the insect will attack ancient oaks that are dominant features of landscapes. Losses of broad-leaved forest stemming from “coniferisation” and clear cutting infested trees are feared (Key 1991). Dead trees are an integral part of an ecosystem, and removal of dead or dying trees to manage *A. biguttatus* would alter the function of the system. Use of insecticides (particularly in urban areas) and biological controls would likely be pursued, just as they were for *A. planipennis* (Haack et al. 2002).

The potential economic impact of *A. biguttatus* in the US is difficult to predict because this species typically occurs in mixed populations with other pests of oak within its native range. The severity of damage varies depending on host availability, stand composition, and forest health, among other factors (Ciesla

2003, Vansteenkiste et al. 2004). Establishment and spread by this insect could jeopardize valuable oak forests, domestic and foreign forest product industries, and the nursery trade. Significant economic losses may result from infestation of live tree hosts or feeding damage that may impact quality of timber, pulp and other forest products (Ciesla 2003). Phloem feeding by this destructive insect can kill a tree or predispose it to further attack by secondary pests (Hartmann and Blank 1992, Blank 1997, Moraal and Hilszczanski 2000a, 2000b, Ciesla 2003, Vansteenkiste et al. 2004).

Control measures could be costly and ineffective given the elusive nature of *A. biguttatus*. Feeding larvae are generally well protected while tunneling inside the host (Ciesla 2003, Vansteenkiste et al. 2004).

Concerns surrounding *A. biguttatus* are heightened by the recent US invasion of a closely related species, emerald ash borer (*Agrilus planipennis*), that has killed thousands of ash trees in Michigan (Haack et al. 2002).

Risks associated with *A. biguttatus* have been evaluated previously. In the Exotic Forest Pest Information System (EXFOR), Ciesla (2003) considered the overall risk posed by the insect to be very high, but this assessment was highly uncertain. The potential for establishment, spread, economic damage, and environmental damage were considered high (Ciesla 2003).

Symptoms

Moraal and Hilszczanski (2000b, 2000a) documented the emergence of hundreds of adults from a single oak trunk, with each specimen leaving a D-shaped exit hole ranging in size from 2-4mm. A heavy infestation has been described as, “38 exit holes per $0.5m^2$ [$5.4ft^2$] of bark” Moraal and Hilszczanski (2000b, Moraal and Hilszczanski 2000a). In addition, zig-zag pattern galleries over 150 cm [59 in] long have been reported.

Plant condition may also suggest the presence of the beetle. Infested trees may have thin crowns, epicormic shoots, or exhibit signs of dieback (Moraal and Hilszczanski 2000b, 2000a).

Known Hosts

Agrilus biguttatus feeds primarily on oak. Reports of *A. biguttatus* feeding on poplar are probably based on misidentifications of *A. ater*.

Hosts	**References**
Fagus sp. (beech)	(Key 1991)
Fagus sylvatica (beech, European)	(Hellrigl 1978, Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003)

Hosts	**References**
Castanea sativa (chestnut, European or sweet)	(Hellrigl 1978, Key 1991, Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003)
Quercus sp. (oak)	(Jacquiot 1976, Foster 1987, Key 1991, Hackett 1995a, 1995b, Moraal and Hilszczanski 2000a, Hilszczanski and Kolk 2001, Ciesla 2003, Denton 2004, Vansteenkiste et al. 2004)
Quercus suber (oak, cork)	(Hellrigl 1978, Moraal and Hilszczanski 2000b, Ciesla 2003)
Quercus pubescens (oak, downy)	(Hellrigl 1978, Moraal and Hilszczanski 2000b, Ciesla 2003)
Quercus petraea (oak, durmast)	(Hellrigl 1978, Hartmann and Blank 1992, Blank 1997, Gutowski and Lugowoj 2000, Moraal and Hilszczanski 2000b, Ciesla 2003, Vansteenkiste et al. 2004)
Quercus robur (oak, English)	(Hellrigl 1978, Hartmann and Blank 1992, Blank 1997, Gutowski and Lugowoj 2000, Moraal and Hilszczanski 2000b, Ciesla 2003, Vansteenkiste et al. 2004)
Quercus cerris (oak, European turkey)	(Hellrigl 1978, Moraal and Hilszczanski 2000b, Ciesla 2003)
Quercus ilex (oak, holly)	(Hellrigl 1978, Moraal and Hilszczanski 2000b, Ciesla 2003)
Quercus rubra (oak, northern red)	(Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003)
Quercus pyrenaica (oak, Pyrenean)	(Echevarria Mayo and Echevarria Leon 1998)
Populus sp. (poplar[1])	(Hellrigl 1978, Moraal and Hilszczanski 2000b, Ciesla 2003)

1. Dubious host record, possibly a misidentification (*A. biguttatus* may have been confused with *Agrilus ater*) (Hellrigl 1978).

Known Distribution

Agrilus biguttatus is present in much of eastern and western Europe, Russian Asia, northern Africa, and the Middle East.

Location	**References**
Algeria	(G. Curletti, personal communication)
Azerbaijan	(Moraal and Hilszczanski 2000b, Ciesla 2003)
Belarus	(Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003)

Location	**References**
Belgium	(Vansteenkiste et al. 2004)
Czech Republic	(Moraal and Hilszczanski 2000b, Ciesla 2003)
England	(Allen 1987, Foster 1987, Godfrey 1987, Allen 1988, Key 1991, Morris 1991, Verdcourt 1992, Else 1994, Smith 1994, Hackett 1995a, 1995b, Jones 1996, Alexander and Foster 1999, Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003, Denton 2004)
France	(Jacquiot 1976, Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003)
Germany	(Hartmann and Blank 1992, Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003, Anon. 2005)
Hungary	(Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003)
Morocco	(Curletti 2005)
Netherlands	(Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003, Moraal 2003)
Iran (Persia)	(Hellrigl 1978)
Poland	(Gutowski and Lugowoj 2000, Moraal and Hilszczanski 2000b, 2000a, Hilszczanski and Kolk 2001, Ciesla 2003)
Russia	(Jacquiot 1976, Hellrigl 1978, Moraal and Hilszczanski 2000a, 2000b, Ciesla 2003)
Spain (Madrid)	(Echevarria Mayo and Echevarria Leon 1998)
Turkey	(Hellrigl 1978)
Ukraine	(Moraal and Hilszczanski 2000b, 2000a, Ciesla 2003)

Potential Distribution within the US

In Europe and Asia, *A. biguttatus* generally occurs in climates ranging from warm and dry to more temperate with adequate rainfall to support forest trees. The currently reported distribution of *A. biguttatus* suggests that the pest may be most closely associated with biomes characterized as: desert and xeric shrublands; Mediterranean scrub; temperate broadleaf and mixed forests; and temperate coniferous forests. Consequently, we estimate that approximately 68% of the continental US would have a climate suitable for establishment of *A. biguttatus*.

Survey

Sampling for *A. biguttatus* will be exceptionally difficult because there are no known baits or traps for this insect. Neither beat- nor sweep-sampling is effective to collect adults (Foster 1987, Allen 1988**.** In Europe, surveys have relied heavily on visual inspection of known hosts, primarily oaks, for D-shaped exit holes (Foster 1987, Hackett 1995a, 1995b, Jones 1996, Denton 2004). These characteristic holes are created as adults emerge from trees.

In previous surveys in Europe, adults have been collected in a haphazard approach, typically with the intent of confirming the presence of the species not quantifying its abundance. Single adults have been painstakingly stalked though the woods (Allen 1988). Anecdotal evidence suggests beetles are most active when in sunlight (Godfrey 1987, Allen 1988, Smith 1994).

Significant research is needed to identify possible attractants for *A. biguttatus*. Limited evidence suggests *A. biguttatus* might be attracted to stressed trees (Moraal and Hilszczanski 2000b, Vansteenkiste et al. 2004). If additional research confirms this observation, a trap-tree method might be developed similar to the approach used for monitoring *A. planipennis*, the Emerald ash borer (USDA 2005). For *A. planipennis*, girdling is used to stress a tree (i.e., the trap tree). Tanglefoot is spread above girdle to trap incoming adults. At the end of the season, trees are felled and bark is stripped to examine for the presence of developing larvae.

This pest could be confused with other indigenous and exotic buprestid species in the US (Ciesla 2003): *A. planipennis*, *A. cuprescens* (=*aurichalceus*), *A. cyanescens*, *A. derasofasciatus*, *A. hyperici*, *A. pilosovittatus*, and *A. sinuatus* (Haack et al. 2002). A dubious host record in the literature reflects the potential difficulty with identification. According to Hellrigl (1978) *A. ater* may have been misidentified and mistaken for *A. biguttatus* on *Populus* sp. (see 'Known Hosts'). *Agrilus biguttatus* has not been reported on *Populus* elsewhere. Conspicuous life stages (pupa and adult) may be positively identified by close examination of morphological characters by a well-trained taxonomist.

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Curculio elephas[*]

Scientific Name
Curculio elephas Gyllenhal

Synonyms:
none known

Common Names
Chestnut weevil

Fig. 1. Adult *Curculio elephas*
[Image from http://galerie-insecte.org/galerie/view.php?adr=.%2Fimage%2Fdos13%2Ftemp%2Felephas_profil.jpg]

Type of Pest
Weevil, seed feeder

Taxonomic Position
Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Coleoptera, **Family:** Curculionidae

Reason for inclusion in manual
Former CAPS Priority Pest (FY 2004)

Pest Description
"Length about 5.2-7.2 mm, measured from apex of pronotum to apex of elytra; entire body rufous to castaneous; except for rostrum, integument densely squamose; scales tan, feebly variegated, not hairlike, not strongly lengthened or elevated along posterior half of suture; body slender; in profile, elytra strongly convex behind basal 1/5; antennal funicle with scales recumbent, second segment much shorter than first, seventh segment longer than basal club segment; scutellum narrow, distinctly elongate; in profile, mesosternum not strongly protuberant anteriorly; metasternum not strongly convex or tuberculate medially; front femur strongly dentate; hind femur with tooth moderately large, posterior edge slightly concave and meeting ventral surface of femur at obtuse angle. Males (distinguished by having first visible sternum of abdomen concave medially): Rostrum about half as long as body, coarsely sculptured, carinate/sulcate from base to antennal insertions; antenna inserted near basal 3/5 of rostrum, scape about as long as basal 6 funicle segments; front femur in profile with scales of ventral margin erect, appearance fringed or shaggy; last visible sternum with pair of apical tufts of setae; pygidium exposed, with large median tuft, without bare median concavity. Females: Rostrum about as long as body, much longer than elytra, straight or feebly curved in basal half; antenna inserted slightly beyond basal 1/4 of rostrum, scape about as long as basal 4-4.5

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[*] This document is largely excerpted from the report: Venette, R.C., E.E. Davis, H. Heisler, and M. Larson. 2003. Mini-Risk Assessment: Chestnut weevil, *Curculio elephas* (Gyllenhal) [Coleoptera: Curculionidae]. Available on-line at www.aphis.usda.gov/ppq/ep/pestdetection/pra/celephaspra.pdf

funicle segments; last visible sternum with apicornedian impression feeble or indistinct, with long, shaggy apical fringe; pygidium exposed apically, with prominent apical fringe, setae less prominent than those of last visible sternum" (Fig. 1) (reviewed in USDA 1983).

"The egg ... is subelliptical, white when deposited, without sculpturing, and measuring 0.40-0.50 x 0.30-0.40 mm" (reviewed in USDA 1983).

"Larvae [have a] maximum length [of] about 9.50 mm. Body white, C-shaped, tapered ... Head nearly circular in outline, convex dorsally; light orange brown except for darker brown mandibles; dorsal setae and sutures...; ocelli indistinct. ... Anal opening distinctly X-shaped" (reviewed in USDA 1983).

"Pupae [have a] maximum length [of] about 9.0 mm. Body white except for eyes, which become dark reddish-brown. ... [Rostrum of] females longer, reaching tips of hind wing pads" (Fig. 2) (reviewed in USDA 1983).

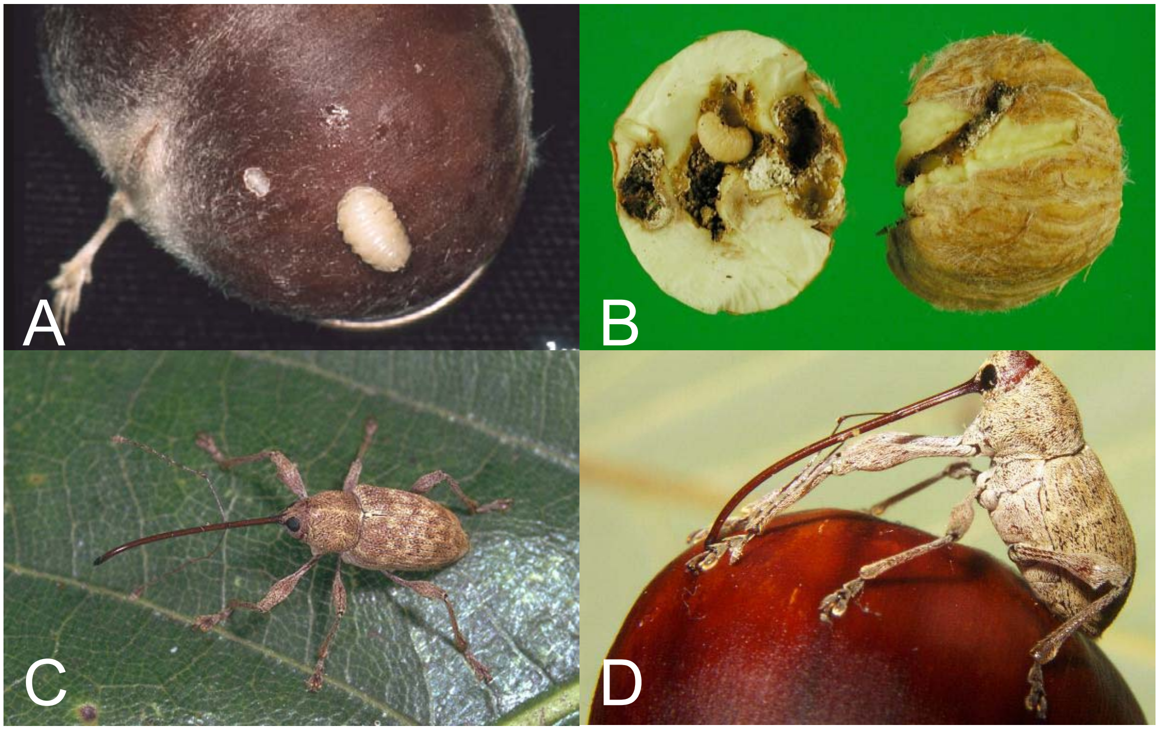

Fig. 2. Life stages of *Curculio elephas*: A) Larva on chestnut; B) Larva and associated damage inside chestnut; C) Adult on chestnut leaf; D) Adult on chestnut.
[Images from: A) Jerry A. Payne, USDA ARS, www.invasive.org; B) http://www.pierroton.inra.fr/IEFC/bdd/patho/patho_affiche.php?langue=en&id_fiche=6; C) http://www.inra.fr/Internet/Produits/HYPPZ/IMAGES/7031480.jpg; D) F. Köhler, http://www.koleopterologie.de/gallery/fhl11/curculio-elephas-foto-koehler.html]

Biology and Ecology

Curculio elephas develops in chestnuts and acorns (Debouzie and Pallen 1987, Speranza 1999, Ploye and Menu 2000, INRA 2003a, Soula and Menu 2003). Adults feed on young nuts (reviewed in USDA 1983). The majority of damage is attributed to larvae. This weevil has a unique life cycle (Fig. 3) (Hrašovec 1993, Speranza 1999). Dispersal and overall mobility are limited and the weevil remains in or near the fruit (i.e., nuts) of its host plant or the host plant itself-throughout its life (Debouzie and Pallen 1987, Menu et al. 2000, Debouzie et al. 2002).

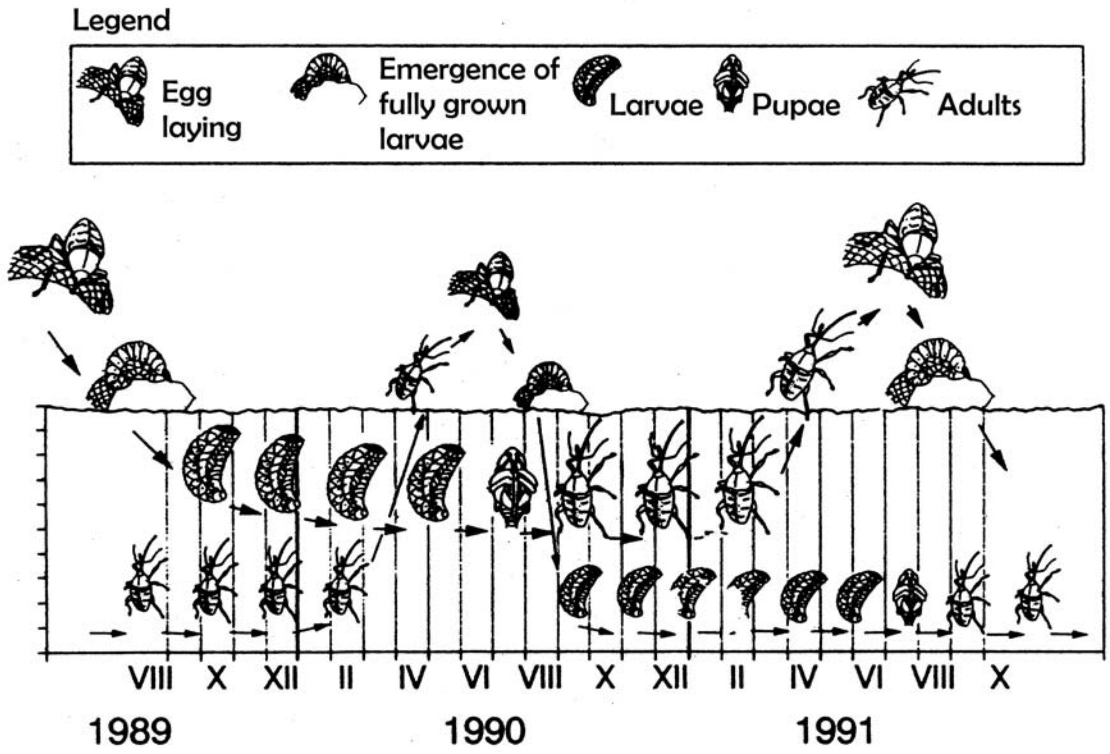

Fig. 3. Population phenology of *Curculio elephas* in Croatia from 1989 to 1991.
[Reproduced from Hrašovec (1993)]

Adults emerge underneath the host tree, fly to nuts, and mate; females then oviposit inside the nuts of the tree under which they emerged (Debouzie and Pallen 1987). Adult emergence can occur between June and November, though emergence from August to September is more typical in much of Europe (USDA 1983, Menu 1993, Menu and Debouzie 1993, Speranza 1999, Menu et al. 2000, Ploye and Menu 2000). The broad window of emergence is partially explained by the range in dates when larvae burrow into soil, between October and November of the preceding year (Speranza 1999). “A newly eclosed adult remains in the overwintering chamber for three to four weeks” (Paparatti and Speranza 1999).

After emergence adults feed for a week (reviewed in INRA 2003a) by puncturing young acorns or chestnuts (reviewed in USDA 1983). Mating and oviposition begin in August and continue until September (Ploye and Menu 2000, Soula and Menu 2003). In the field, adult females lived an average of 9.5 days while in protected laboratory conditions female lifespan increased to 21 to 28 days (Desouhant 1996, Debouzie et al. 2002). The female is active throughout the oviposition period, which can last several weeks (reviewed in USDA 1983). Oviposition takes place on mature nuts or acorns (reviewed in USDA 1983). Females do not select nuts for oviposition based on the size of the nut, nor does the presence of other eggs or larvae of the species deter oviposition (Desouhant 1998).

Eggs are most often laid singly within holes in the chestnut, not on the leaves or branches of the tree (USDA 1983, Ploye and Menu 2000, Soula and Menu 2003). Females puncture the nut with their rostrum and deposit eggs into the hole (Speranza 1999). Multiple eggs may be inserted into a single nut. A third of the time, females oviposit in previously bored holes, thus saving energy and time (Desouhant 1996). Each female can lay 20-50 eggs, with an average of 43 eggs (USDA 1983, Ploye and Menu 2000, INRA 2003a). Females can lay an average of 1.9 eggs each day for the first 10 days after emergence (Ploye and Menu 2000).

Larvae complete development inside the nut. There are four instars (Ploye and Menu 2000). Typically, one or two larvae will be located in each nut, although as many as 19 larvae have come from a single nut (USDA 1983, Paparatti and Speranza 1999, Ploye and Menu 2000, Debouzie et al. 2002). Larval development is completed inside same fruit where eggs were deposited (Menu et al. 2000, Ploye and Menu 2000, Soula and Menu 2003). Late instar larvae [ca. 4-10 weeks after oviposition (reviewed in USDA 1983)] emerge from the nut or wait until the nut drops to the ground, then they burrow into soil under the tree to diapause (reviewed in USDA 1983, Debouzie and Pallen 1987). To leave the nut, the insect must chew an exit hole. Once out of the nut and on the ground, larvae bury themselves in a small cell where they spend the winter in diapause (USDA 1983, Manel and Debouzie 1997, Menu et al. 2000, Soula and Menu 2003). Larvae enter diapause between October and December (Speranza 1999) to survive challenging environmental conditions (Menu 1993). Further larval development begins in March and continues until adults emerge. Some larvae can delay development and remain in the soil for more than one year (Paparatti and Speranza 1999, Speranza 1999).

Most larvae pupate the year following oviposition but some of this cohort may pupate after 2-3 years (reviewed in USDA 1983, Soula and Menu 2003). After one year in diapause, 59% of larvae emerge as adults, but fewer (37%) emerge after 2 years in an extended diapause; very few (4%) emerge after 3 years in the ground (Menu et al. 2000, Soula and Menu 2003).

Pupation typically begins in May but can start in August (reviewed in USDA 1983). In France, pupation occurs in July and August with adult emergence occurring between August and the first part of October (Soula and Menu 2003). Pupation generally occurs outside of the nut in the soil (reviewed in USDA 1983).

Environmental conditions during the pre-diapause period influence the length of the life cycle and whether or not diapause is induced (Manel and Debouzie 1997). Several studies describe developmental thresholds and accumulated degree days [˚C] necessary for the completion of each phenological stage (Table 1). However, because of the complexity of the life-cycle, very few models have been developed to describe completion of the life-cycle development in response to temperature.

Table 1. Developmental threshold and degree day requirements for *Curculio elephas*

Stage	Developmental threshold (˚C)	Degree Days (˚C)	Notes	Reference
Egg	6.5	108.9	Estimated under natural conditions	(Manel and Debouzie 1997)
Larva	0	103 82 114 311 (for 1st 3 stages)	Stage 1 Stage 2 Stage 3 Stage 4	(Manel and Debouzie 1997)
	0	593	Median time for larval development	(Manel and Debouzie 1995)

Drought conditions lead to hard-packed soils that inhibit the emergence of the adults from the ground (Menu and Debouzie 1993, Soula and Menu 2003). Such soils can prevent the emergence and reproduction of 27-87% of females (Menu and Debouzie 1993). Wet conditions can also adversely affect populations. High soil moisture can increase mortality (Önuçar and Ulu 1989). Much of the mortality associated with moist soils may be attributed to microbial pathogens. Rainy conditions can affect oviposition success (Debouzie et al. 2002).

Survival rates during the larval stage are generally low (Menu 1993). Predation from small animals (including millipedes), infection by pathogens, and hard or frozen soil (preventing larvae from forming pupation cells) contribute significantly to overall mortality rates (Menu 1993, Soula and Menu 2003). Mortality is particularly affected during the prediapause stage when humans collect large numbers of chestnuts for consumption (Soula and Menu 2003). Parasite attack in *C. elephas* is minimal (Debouzie et al. 2002).

Pest Importance

Curculio elephas is an oligophagous pest, attacking several species of chestnut (*Castanea* spp.) and oak (*Quercus* spp.). The insect is considered a somewhat specialized, but still economically important pest (Bürgés and Gál 1981, Debouzie et al. 1996, Branco et al. 2001). This weevil is one of the most serious pests of European chestnut (*Castanea sativa*) in Europe; damage is caused by adults feeding on the base of young nuts and larvae feeding on mature nuts (USDA 1983). Reports of significant damage come from Algeria (Chakali et al. 2002), Italy (Paparatti and Speranza 1999), Poland (Pomorski and Tarnawski 1980), Portugal (Menu 1993, Branco et al. 2001), Spain, and Turkey (Canakcioglu 1969, Yaman et al. 1999). Impacts to chestnut depend on chestnut variety (INRA 2003a). Adult feeding can cause up to 20% premature nut drop while combined larval and adult feeding can cause up to 90% crop loss (USDA 1983, Paparatti and Speranza 1999, INRA 2003a, 2003b). Larval feeding damage on acorns can have a negative effect on seedling vigor (Branco et al. 2001). The percentage of attacked acorns in Portugal is estimated between 50 – 63%, of which 89 – 95% of the damage causing pest population is *C. elephas* (Branco et al. 2002). Although infested acorns are still able to germinate and survive, there are few reserves left for seedling growth (Branco et al. 2002). Thus, the pest may impede regeneration of oak and chestnut stands.

We also speculate that *C. elephas* may adversely the health of chestnut by interacting with *Endothia parasitica*, the causal agent of chestnut blight. Although there are no reports of *C. elephas* vectoring the pathogen or predisposing trees to infection, Russin et al. (1984) noted that ~42% of native curculionids (i.e., *Acoptus suturalis* and *Rhyncolus brunneus*) carried the pathogenic fungus. Pakaluk and Anagnostakis (1997) conjecture that *A. suturalis* may vector the pathogen.

Symptoms

Trees exhibit few symptoms when infested with *C. elephas*. Holes created by females for oviposition are not easily seen (reviewed in USDA 1983). Infested nuts may drop prematurely. The meat within the nut will be consumed and discolored, but the weevil is generally not considered detectable in the field unless the nut is cracked open.

Known Hosts

Larvae of *Curculio elephas* feed and develop within chestnuts (*Castanea* spp.) and acorns (*Quercus* spp.) (Debouzie et al. 1996, Ploye and Menu 2000, INRA 2003a).

Hosts	References
Castanea sativa (European chestnut)	(CAB 2000; Debouzie 2002)
Castanea vesca (sweet chestnut)	(Canakcioglu 1969)
Quercus suber (cork oak)	(Branco 2001; CAB 2000)
Quercus robur (pedunculate oak)	(CAB 2000)

Known Distribution

This weevil is generally distributed in Europe and portions of North Africa and the Middle East (USDA 1983).

Location	**References**
Austria	(CAB 2000)
Algeria	(CAB 2000)
Bulgaria	(CAB 2000)
Croatia	(CAB 2000)
Cyprus	(CAB 2000)
Czechoslovakia	(CAB 2000)
France	(CAB 2000)
Germany	(CAB 2000)
Greece	(CAB 2000)
Hungary	(CAB 2000)
Israel	(CAB 2000)
Italy	(CAB 2000)
Poland	(CAB 2000)
Portugal	(CAB 2000)
Russia	(CAB 2000)
Spain	(CAB 2000)
Switzerland	(CAB 2000)
Tunisia	(CAB 2000)
Turkey	(CAB 2000)
Yugoslavia	(CAB 2000)

Potential Distribution within the US

Curculio elephas is currently found in the eastern Palearctic (reviewed in USDA 1983). This region is generally characterized by a temperate climate (reviewed in CAB 2000). The currently reported global distribution of *C. elephas* suggests that the pest may be most closely associated with temperate broadleaf and mixed forests and temperate coniferous forests. Based on the distribution of climate zones in the US, we estimate that approximately 47% of the continental US would be suitable for *C. elephas*.

Survey

Surveys for *C. elephas* must typically depend on emergence traps or visual inspection of potentially infested chestnuts or acorns. During visual inspections, nuts must be opened to reliably detect larvae (reviewed in USDA 1983). Because of difficulties in reliably identifying immature stages of the insect, any curculionid identified during a survey of chestnuts or acorns should be reared to an adult (reviewed in USDA 1983).

Emergence traps may also be used to collect newly emerged adults (Hrašovec 1993). In one case, the emergence trap was an inverted 1 m^2 [~10 ft^2] wooden box; holes in the side of each box provided light to which the insects were attracted (Menu 1993). Adults were captured in small cages attached to the box by a plastic tube.

No sex or aggregation pheromone has yet been identified for *Curculio elephas*. However, “male-specific compounds” that attract females and some males have been identified for the pecan weevil, *Curculio caryae* (Bartlelt 1999). The aggregation pheromone of *C. caryae* is a five component blend of grandisol and isomers of octodenol (Hedin et al. 1997). Four of these compounds are also used by boll weevil (Bartlelt 1999). No work has been conducted to develop or adapt a pheromone-baited trap for *C. elephas*.

In the US, larvae and adults of *C. elephas* are most likely to be confused with other *Curculio* spp. that also feed on chestnuts. Two native or naturalized *Curculio* spp., *C. caryatrypes* and *C. sayi*, may also affect chestnuts, but adults of these taxa are easily distinguished from *C. elephas*. In addition, other weevils of the genus *Conotrachelus* (*C. carnifer* and *C. posticatus*) may develop in chestnut and complicate surveys for *C. elephas*. From Europe, taxa that might be confused with *C. elephas* include *C. nucum* and *C. glandium*, though the latter species is more commonly associated with acorns (reviewed in USDA 1983). No taxonomic keys are available to identify curculionid pupae to genus or species, and distinguishing features of many immature *Curculio* spp. have not been described (reviewed in USDA 1983).

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Platypus quercivorus[*]

Fig. 1. Adult *Platypus quercivorus*. [Image from http://www.zin.ru/animalia/coleoptera/images/foto/platypus_cf_quercivorus,_fe.jpg]

Scientific Name

Platypus quercivorus Murayama

Synonyms:
Crossotarsus quercivorus Murayama, 1925
Crossotarsus sexfenestratus, 1937

Common Names

Ambrosia beetle

Type of Pest

Wood borer, ambrosia beetle

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Coleoptera, **Family:** Platypodidae

Reason for inclusion in manual

CAPS Priority Pest (FY 2007)

Pest Description

Platypus [=*Crossotarsus*] *quercivorus* was first described by Murayama in 1925 (Murayama 1925). A subsequent morphological description was published by Beeson (1937) and in 1972 the genus was reassigned to *Platypus* by Schedl (1972).

“[Coloration] Ferruginous brown, the head and the apex of elytra darker, underside yellowish brown” (Murayama 1925).

“[Male]. Head with front flat, covered with an irregular rugose reticulation, a short depressed median line between the bases of the antennae; vertex rather abruptly separated from the front, with a narrow black median line, sparse rugose punctures, and long aureous hair” (Murayama 1925).

“Elytra elongate, with sides parallel in the anterior two thirds and gradually diminished about one third of the breadth towards the apex; upper surface with a slight declivity in the posterior third, with the apex abruptly truncated. ... Underside with scanty long yellow hair and large porelike punctures, abdominal

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[*] This document is largely excerpted from the report: Davis, E.E., S. French, and R.C. Venette. 2005. Mini-Risk Assessment: Ambrosia Beetle, *Platypus quercivorus* Murayama [Coleoptera: Platypodidae]. Available on-line at www.aphis.usda.gov/ppq/ep/pestdetection/pra/pquercivoruspra.pdf

segments convex, the 7th with a large transversal shallow oval depression" (Murayama 1925).

Fig. 2. *Platypus quercivorus* adult (left) and wood boring damage (right; images not to scale). [Image from http://ss.ffpri.affrc.go.jp/research/ryoiki/07for-entom/07.html].

"[Female]. Front and vertex the same as in the male. Prothorax subquadrate, shining, with fine punctures and median sulcus, as in the male, on each side of the sulcus with 3-5 large round touched depressions in two rows, each depression being surrounded by a black bar. ... Elytra as in the male, excepting in the more gently rounded sides and declivity ... Underside a little paler than in the male, with stronger convexity on each abdominal segment" (Murayama 1925).

Table 1. Mean size of male and female *Platypus quercivorus*

	Male	Female
Length	4.46 mm	4.54 mm
Length of prothorax	1.29 mm	1.33 mm
Breadth of prothorax	1.15 mm	1.08 mm
Length of elytra	2.38 mm	2.42 mm
Breadth of elytra (at the base)	1.15 mm	1.15 mm

Other factors may help distinguish *Platypus* spp. Certain symbiotic fungi are sufficiently species specific that identification of the fungus may assist with identification of the beetle (Baker 1963, Batra 1963). *Platypus quercivorus* is closely associated with *Raffaelea quercivora* (see chapter in this reference). Wood-boring damage may also differentiate *Platypus* spp. For example, *P. quercivorus* produces splinters that accumulate with expelled frass near the base of infested trees during gallery construction; a fine sawdust is characteristic of other related species (Kuroda and Yamada 1996, Ciesla 2003, reviewed in CAB 2004).

Biology and Ecology

In Japan, *Platypus quercivorus* is typically a univoltine species; however, adults may emerge in late spring and autumn of the same year (Kinuura 1995, Soné et al. 1998, Kinuura 2002). New adults emerge and disperse beginning in late June through early October or November (Soné et al. 1998, Kinuura 2002). Adult males first colonize new hosts and trigger a mass attack; possible attractants include plant volatiles from wounded hosts, aggregation pheromones, and sound released by male beetles (Ohya and Kinuura 2001, Kinuura 2002, Kobayashi and Ueda 2003, Atkinson 2004). Males typically enter standing trees on the lower portion of the trunk, within about a meter of the ground (Hijii et al. 1991, Igeta et al. 2004). Larger trees (16-40 cm diameter at breast height (dbh) [6-16 in]) are preferred over smaller trees (9.5-16 cm dbh [4-6 in]) (Soné et al. 1995). Although the density and position of entry holes may be affected by several factors, males create entrance holes where tree diameter and moisture content are optimal (Soné et al. 2000, Esaki et al. 2004, Igeta et al. 2004).

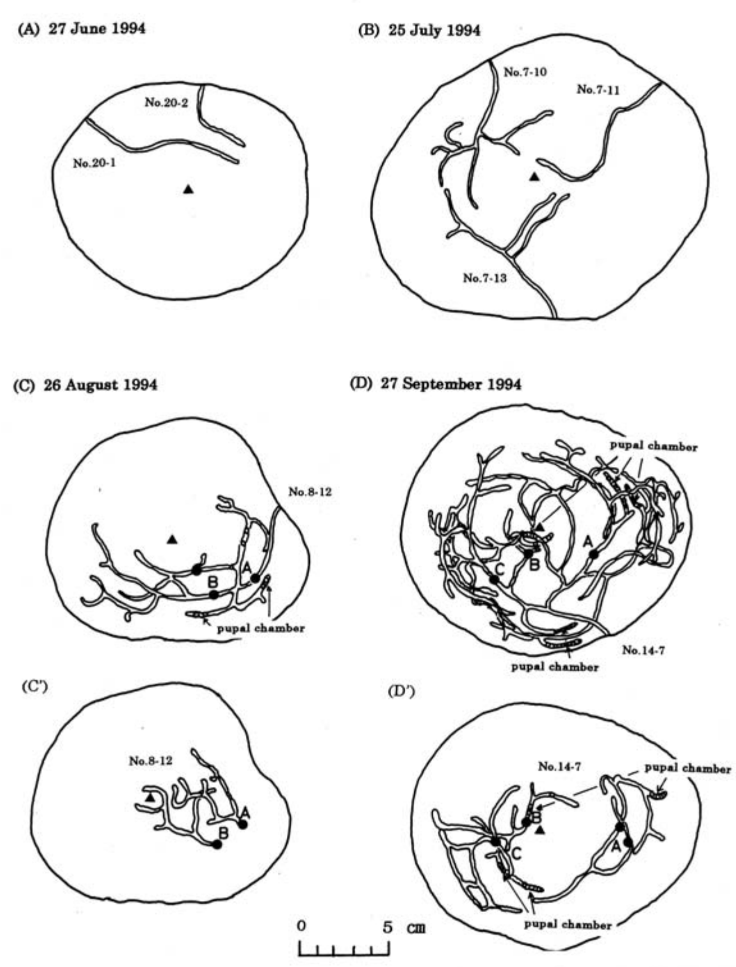

Fig. 3. Gallery construction by *P. quercivorus* in logs.
[Image reproduced from Soné et al. (1998).]

A male will bore into a tree horizontally to create a mating gallery. A female will join the male at the entrance, mate and initiate construction of an oviposition gallery (Fig. 2, Soné et al. 1998). The oviposition gallery is branched several times laterally and vertically to allow developing larvae to bore extensively throughout a tree (Kinuura 2002). Well-developed gallery systems over 387 cm [152 in] in length with up to seven layers of horizontal tunnels have been described (Soné et al. 1998). The female carries fungi in a mycangia, specialized integumentary pores in the pronotum designed to carry fungal spores or fragments. Fungi distributed in the oviposition chamber are cultivated to provide a food source. Eggs are laid in individual notches at terminal ends within the horizontal gallery walls about 2-3 weeks after gallery construction is initiated (Kinuura 2002, Ciesla 2003). As the female creates the oviposition gallery, she will carry debris to the male who remains at the entrance hole, presumably to protect against predators (Soné et al. 1998). The males will expel frass and splintered wood from the gallery system. Adult beetles are monogamous and remain in the gallery until their brood fully develops; afterwards, the adults die (Soné et al. 1998).

Egg hatch occurs about one week after oviposition (Kinuura 1995, Esaki et al. 2004). Larvae feed on the fungi lining the gallery wall (Kinuura 2002). There are typically 5 instars. Depending on the season, final instar larvae may or may not enter "hibernation" and overwinter in the larval gallery (Soné et al. 1998). Fifth instars pupate in 1 cm-long pupal chambers within the larval gallery (Soné et al. 1998, Kinuura 2002). Pupae are larger than adults and white (Kinuura 1995). Gender can be differentiated in this stage (Kinuura 1995). The beetle may overwinter as a pupa or adult; however, adults that remain in the host until the following spring may not survive (Soné et al. 1998).

Reproductive success of *P. quercivorus* is reportedly higher in fresh logs than in living trees (Soné et al. 1998). Fewer adults emerged from living trees (3.5-9.7 adults) than logs (40-60 adults) (Sato and Arai 1993, Soné et al. 1998, Ciesla 2003, reviewed in CAB 2004). As many as 161 individuals may inhabit a gallery, and an average of 20-30 new adults may emerge from a gallery in summer and spring, respectively (Soné et al. 1998).

Platypus quercivorus is the only known vector for the ambrosia fungus *Raffaelea quercivora* in oaks (Kinuura 2002, Kubono and Ito 2002, Ito et al. 2003b, Ito et al. 2003a). Though the exact mechanism of host inoculation is not known (e.g., active or passive, or a combination), *R. quercivora* is introduced by adult *P. quercivorus* beetles as they bore into new hosts (Kinuura 2002). All life stages of *P. quercivorus* can use this particular fungus as a food source (Baker 1963, Cooke 1977, Kinuura 2002). *Raffaelea quercivora* has been isolated from necrotic tissue of inner bark, stained sapwood and heartwood; body surfaces of male and female *P. quercivorus*; the proventriculus or terminal foregut of males and females; the mycangia of females; and gallery systems constructed by adults prior to emergence (Ito et al. 1998, Kinuura 2002, Ito et al. 2003b, Ito et al. 2003a, reviewed in CAB 2004).

Pest Importance

Platypus quercivorus is an important pest of Japanese oaks, chestnuts and other Fagaceae. *Raffaelea quercivora*, an ambrosia fungus vectored by *P. quercivorus*, appears to be the causal agent of Japanese oak disease (also known as Japanese oak wilt). In Japan, the *R. quercivora* - *P. quercivorus* complex has killed approximately 100,000-200,000 fagaceous trees annually since about 1980; the majority of affected hosts are *Quercus serrata* and *Q. mongolica* var. *grosseserrata* (Ito et al. 2003b, Ito et al. 2003a). This is the first time that an ambrosia beetle-fungus complex has killed healthy trees (Kamata 2002, Kamata et al. 2002, Ito et al. 2003b, Ito et al. 2003a, Esaki et al. 2004). Tree death can occur the same year as a mass attack by *P. quercivorus*, but most oaks die within three years (Kamata 2002, Kubono and Ito 2002, Kobayashi and Ueda 2003). Wilting may be evident within 10 days (Ito et al. 2003b, Ito et al. 2003a). Generally, the white oak group seems highly susceptible, but species of white oaks show differing degrees of susceptibility.

Platypus quercivorus can slow growth and increase mortality of host and non-host trees. Newly felled trees and cut timber contribute to mass attack and death of nearby living trees (Kobayashi and Hagita 2000, Igeta et al. 2004). *Platypus quercivorus* typically will bore into trees adjacent to areas of mass attack, even nutritionally unsuitable hosts (A. Ueda, personal communication, Ueda and Kobayashi 2001). Wood boring predisposes trees to further damage by secondary pests including other ambrosia and bark beetles, decay fungi and other microorganisms (Beaver 1989, Kozlowski et al. 1991). This type of damage impacts wood quality, both aesthetically (discoloration of sapwood) and structurally (Hijii et al. 1991, Manion 1991, Kuroda and Yamada 1996, Ito et al. 1998, Yamato et al. 2001, Ito et al. 2003b, Ito et al. 2003a).

Extensive oak mortality in Japan may have impacted habitat for Asian black bears, driving bears into more populated areas where numerous humans were attacked (Yamazaki 2004). To prevent further attacks, numerous bears were killed, 170 in one district alone (Yamazaki 2004).

The economic impact of *P. quercivorus* by itself in Japan is difficult to measure, especially because it occurs in mixed populations with other secondary attackers (Inoue et al. 1998, Soné et al. 1998, Ueda and Kobayashi 2001). Damage associated with *P. quercivorus* was reported ca. 70 years ago, but it is not clear if this damage referred to tree mortality or whether damage was caused by the beetle alone or the beetle and *R. quercivora* (reviewed in Hamaguchi and Goto 2003). In Japan, mortality from *P. quercivorus* was less severe in broadleaf evergreen forests than in broadleaf deciduous forests (M. Yamato, personal communication, Zhou undated).

The risks posed by *P. quercivorus* to natural resources in the US have been evaluated previously. Ciesla (2003) concluded that the insect poses very high risk but recognized that this rating was very uncertain. Uncertainty stemmed from a significant lack of knowledge about how the insect and pathogen might affect oak species that occur in the US. If *Quercus* spp. in the US are susceptible to attack by *P. quercivorus* and infection by *R. quercivora*, the economic impact from yield reductions, quality losses and trade restrictions could be significant. Establishment and spread of the pest complex could jeopardize domestic and international trade in wood products and ornamental plants. *Raffaelea quercivora* is listed on the EPPO Alert List (EPPO 2005). There is no known control for the disease.

Symptoms

In Asia, stands suspected of being infested with *P. quercivorus* often have wilted canopies during the summer in the absence of drought and/or a reddish-brown discoloration of leaves (Ciesla 2003). Infestation may trigger the formation of tyloses in the tree (Manion 1991, Farrell et al. 2001). As tyloses form, membranes and parenchyma cells expand into xylem vessels and impede water uptake. This may account for the rapid wilting (within one growth season or the same year of pest attack) of susceptible hosts, particularly those belonging to the white oak group which conduct water primarily in the current year's growth. Tyloses occur naturally in areas of older growth (sapwood) where water conductance no longer takes place. Numerous chemical compounds are produced by the tree during the formation of tyloses which serve to protect the tree from decay fungi. In contrast to ring porous white oaks, evergreen oaks are semi-ring- to diffuse-porous trees in which water transport takes place in a number of vessels, not limited to areas of new growth. Pest infestation may not cause wilting as quickly or extensively as in more porous oak species (Agrios 1988, Manion 1991).

In infested stands, splinter-like wood shavings may be present at the base of a tree (Ciesla 2003, reviewed in CAB 2004). Entrance holes and wood shavings produced by males as they create galleries for mating are strong evidence for the presence of the beetle (Kobayashi and Ueda 2002).

Known Hosts

True hosts of *P. quercivorus* are members of the family Fagaceae. At this time, only a few known hosts of *P. quercivorus* occur in the US and all are introduced species: Mongolian oak, sawtooth oak, and Japanese chestnut. Of these, only Mongolian oak is also a known host of the pathogenic fungus *R. quercivora* which is vectored by the beetle.

Hosts	**References**
Castanea crenata (chestnut, Japanese)[2]	(Igeta et al. 2003)
Castanopsis cuspidata (chinkapin, Japanese)	(Wood and Bright 1992, Mori et al. 1995, Ciesla 2003, CAB 2004)
Castanopsis sieboldii (=*C. cuspidate* var. *sieboldii*) (sudajii)	(Kamata et al. 2002, Esaki et al. 2004)
Lithocarpus edulis (=*Pasania edulis*) (oak, Japanese tanbark)	(Murayama 1925, Wood and Bright 1992, Mori et al. 1995, Soné et al. 1995, Soné et al. 1998, Soné et al. 2000, Ciesla 2003, Sato 2003, CAB 2004, Kitajima and Goto 2004)
Lithocarpus glaber (=*Pasania glabra*) (oak, Japanese)	(Wood and Bright 1992, Ciesla 2003, CAB 2004)
Quercus sp. (oak)	(Beaver 1989, Hijii et al. 1991, Soné et al. 1998, Saito et al. 2001, Kobayashi and Ueda 2002, Ito et al. 2003a, Ueda and Kobayashi 2004)

Hosts	**References**
Quercus acuta (oak, Japanese evergreen)	(Wood and Bright 1992, Mori et al. 1995, Kamata et al. 2002, Ciesla 2003, Igeta et al. 2003, CAB 2004, Esaki et al. 2004, Igeta et al. 2004)
Quercus acutissima (oak, sawtooth)[2]	(Wood and Bright 1992, Ciesla 2003, CAB 2004)
Quercus crispuloserrata	(Hijii et al. 1991)
Quercus gilva (ichiigashi)	(Murayama 1925, Wood and Bright 1992, Ciesla 2003, CAB 2004)
Quercus glauca (=*Q. myrsinifolia*) (oak, ring-cup or Japanese blue)	(Murayama 1925, Wood and Bright 1992, Ciesla 2003, CAB 2004)
Quercus mongolica (=*Q. crispula* (=*Q. mongolica* var. *grosseserrata*)) (oak, Mongolian)[2]	(Wood and Bright 1992, Kuroda and Yamada 1996, Mizobuti et al. 1996, Ito et al. 1998, Kinuura et al. 1998, Masuya et al. 1998, Kobayashi et al. 2001, Kuroda 2001, Ohya and Kinuura 2001, Ueda and Kobayashi 2001, Yamato et al. 2001, Kamata et al. 2002, Kobayashi and Ueda 2002, Kubono and Ito 2002, Ciesla 2003, Igeta et al. 2003, Kobayashi and Ueda 2003, CAB 2004, Esaki et al. 2004, Kitajima and Goto 2004, Ueda and Kobayashi 2004)
Quercus myrsinaefolia (oak, Japanese white)	(Wood and Bright 1992)
Quercus phillyraeoides (oak, ubame)	(Ciesla 2003, CAB 2004)
Quercus salicina (urajirogashi)	(Wood and Bright 1992, Mori et al. 1995, Ciesla 2003, CAB 2004, Igeta et al. 2004)
Quercus senata[1]	(Igeta et al. 2004)
Quercus serrata (oak, Konara)	(Hijii et al. 1991, Wood and Bright 1992, Kuroda and Yamada 1996, Ito et al. 1998, Kinuura et al. 1998, Kobayashi et al. 2001, Kuroda 2001, Ohya and Kinuura 2001, Ueda and Kobayashi 2001, Yamato et al. 2001, Kamata et al. 2002, Kobayashi and Ueda 2002, Kubono and Ito 2002, Ciesla 2003, Igeta et al. 2003, Kobayashi and Ueda 2003, CAB 2004, Esaki et al. 2004, Kitajima and Goto 2004, Ueda and Kobayashi 2004)

Hosts	References
Quercus sessilifolia (tsukubanegashi)	(Wood and Bright 1992, Ciesla 2003, CAB 2004)

1. Probable misspelling for *Quercus serrata*. *Quercus* "*senata*" is not a recognized species.
2. These plants occur in the U.S.; introduced species (USDA NRCS 2004).

Although *P. quercivorus* has reportedly initiated galleries on several non-fagaceous species adjacent to an area of mass attack, the insect cannot successfully reproduce on such trees (Dr. Akira Ueda, Hokkaido Research Center, personal communication). Trees that may be attacked but will not support reproduction include:

Cupressaceae [=Taxodiaceae]
Japanese cedar, *Cryptomeria japonica* (Wood and Bright 1992, Ciesla 2003, CAB 2004)

Aquifoliaceae
Chinese holly, *Ilex chinensis* (Wood and Bright 1992, Ciesla 2003, CAB 2004)

Lauraceae
Japanese silver tree, *Neolitsea sericea* (Soné et al. 1995)
common machilus, *Persea* (=*Machilus*) *thunbergii* (Soné et al. 1995, Sato 2003)
wild machilus, *Persea* (=*Machilus*) *japonica* (Soné et al. 1995)
spicebush, *Lindera erythrocarpa* (Wood and Bright 1992, Ciesla 2003, CAB 2004)

Rosaceae
Korean mountain ash, *Sorbus alnifolia* (Kobayashi and Ueda 2002)
Prunus sp. (Wood and Bright 1992, Ciesla 2003, CAB 2004)

Known Distribution

Platypus quercivorus occurs in east and southeast Asia. It is not currently known to occur in the United States. *Platypus quercivorus* is common in parts of Japan and is present, but to a very limited extent, in India, Taiwan, Indonesia, and Papua New Guinea.

Location	References
China[1]	(CAB 2004)
India	(Beeson 1937, Schedl 1972, Beaver 1989, Wood and Bright 1992, Kobayashi and Ueda 2002, Ciesla 2003, CAB 2004)
Indonesia (Java)	(Beeson 1937, Schedl 1972, Beaver 1989, Wood and Bright 1992, Kobayashi and Ueda 2002, Ciesla 2003, CAB 2004)

Location	References
Japan	(Murayama 1925, Schedl 1972, Beaver 1989, Hijii et al. 1991, Wood and Bright 1992, Mori et al. 1995, Soné et al. 1995, Mizobuti et al. 1996, Ito et al. 1998, Kinuura et al. 1998, Masuya et al. 1998, Soné et al. 1998, Kuroda 2001, Ohya and Kinuura 2001, Saito et al. 2001, Esaki et al. 2002, Kamata et al. 2002, Kobayashi and Ueda 2002, Kubono and Ito 2002, Ciesla 2003, Igeta et al. 2003, Kobayashi and Ueda 2003, Sato 2003, CAB 2004, Esaki et al. 2004, Igeta et al. 2004, Kitajima and Goto 2004, Ueda and Kobayashi 2004)
Papua New Guinea	(Schedl 1972, Beaver 1989, Wood and Bright 1992, Kobayashi and Ueda 2002, Ciesla 2003, CAB 2004)
Southeast Asia	(Kamata et al. 2002)
Taiwan (formerly Formosa)	(Beeson 1937, Schedl 1972, Beaver 1989, Wood and Bright 1992, Kamata et al. 2002, Kobayashi and Ueda 2002, Ciesla 2003, CAB 2004)

1. Questionable record. *P. quercivorus* is reportedly present in Taiwan, but not throughout China.
2. Pathogen *Raffaelea quercivora* identified in association with vector *Platypus quercivorus* and oak mortality.
3. Oak mortality associated with *P. quercivorus* and presence of *R. quercivora* is presumed but not isolated.

Potential Distribution within the US

In general, *P. quercivorus* occurs in temperate or tropical climates with adequate seasonal rainfall to support deciduous tree hosts. The currently reported distribution of *P. quercivorus* suggests that the pest may be most closely associated with biomes characterized as temperate broadleaf and mixed forests and tropical and subtropical moist broadleaf forests. Consequently, we estimate that approximately 29% of the continental US would have a suitable climate for *P. quercivorus*.

The climatic tolerances of the beetle may be wider than those of the pathogen. Although the beetle has been reported from a number of countries, the pathogen has only been reported from Japan (reviewed in Kromroy and Venette 2005). Specific abiotic conditions may be needed for the beetle-fungus complex to cause extensive mortality, but these conditions have not been specified.

Survey

Surveys for *Platypus quercivorus* are likely to be difficult. Although an aggregation pheromone has been suggested for another *Platypus* sp. (Milligan et al. 1988), *Platypus*-attractants are not yet sufficiently reliable for use with traps. *Platypus quercivorus* may have a weak attraction to ethanol (Kobayashi and Hagita 2000), but the utility of ethanol for trapping *P. quercivorus* has been questioned (as reviewed in Esaki et al. 2002).

Visual surveys are common for the beetle in declining or suspect stands. Suspect stands have wilted canopies during the summer in the absence of drought and/or a reddish-brown discoloration of leaves (Ciesla 2003). In these stands, oaks are examined for splinter-like wood shavings at the base of a tree (Ciesla 2003, reviewed in CAB 2004) or entrance holes produced by male beetles. Wood shavings (Fig. 3) are produced by males as they create galleries for mating (Kobayashi and Ueda 2002).

Fig. 4. Wood shavings produced by ambrosia beetles (Platypodidae).
[Image from Randy Cyr, www.forestpests.org]

In Japan, Esaki et al. (2004) examined ~46-160 trees/acre for research purposes. Larger trees are more likely to be attacked and entrance holes will be common in the lower trunk of the tree (see 'Biology and Ecology').

For visual surveys, the number of samples needed to detect *P. quercivorus* depends on the frequency of infested trees in a stand and the desired confidence of detecting the beetle when it is present. In areas of Japan with well established pockets of oak wilt, *P. quercivorus* may attack 7-93% of trees that are susceptible to the fungus (Soné et al. 1995, Esaki et al. 2004). For early detection, it would be desirable to detect beetles before they infest this many trees. Assuming that (i) visual inspection of a single tree will locate beetles if they are present on that tree, (ii) a stand has a large number (e.g., >1000) of trees that may be fed upon by *P. quercivorus*, and (iii) trees are selected at random for inspection, binomial statistics can be used to determine the number of trees that must be examined to achieve a desired probability of finding at least one infested tree within a stand when the beetle is present. Figure 4 illustrates how the number of required samples changes as the proportion of trees with *P. quercivorus* and/or the desired probability of detecting at least one infested tree changes. In general, more samples are required as the desired probability of detection increases and as the proportion of trees with beetles decreases (i.e., the insects become rarer in the environment).

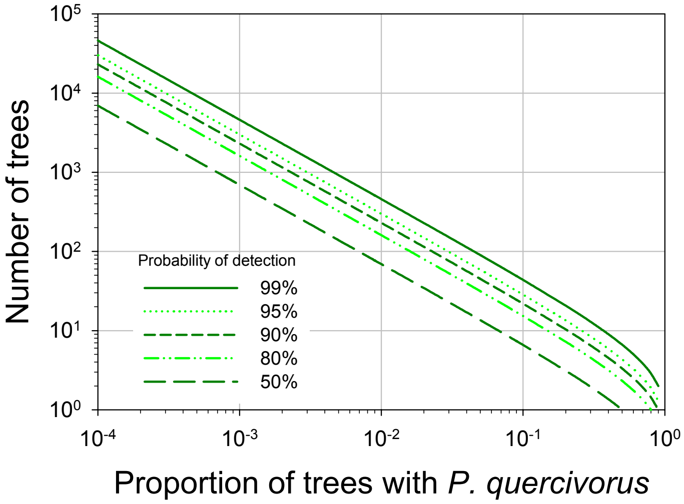

Fig. 4. Required number of trees to be inspected for detection of *P quercivorus* in relation to the proportion of infested trees and the desired probability of detecting this insect. This figure assumes random sampling from a large environment.

As a complement to visual inspections, Japanese researchers recommend the use of interception traps, which non-selectively capture flying insects (Kinuura 1995, Esaki et al. 2002). An interception trap based on the use of nylon screen covered with a sticky coating was light, durable, and effective (Esaki et al. 2002, Esaki et al. 2004). Traps were constructed from a 1-m^2 [~10 ¾ ft^2] piece of nylon mesh (mesh size = 2.4x2.6mm). Wooden stakes were attached horizontally to the top and bottom of the panel. The top stake was used to hang the trap, and the bottom stake provided weight to keep the trap straight. Traps were hung so that the bottom stake was 0.5 m [~1 ½ ft] from the ground. This places the trap in the zone where most adults are captured (Hijii et al. 1991, Kobayashi and Hagita 2000, Ueda and Kobayashi 2001, Igeta et al. 2004). This trap design is particularly convenient in areas with steep terrain or high winds.

Traps should be placed in June or July, the start of adult flight in Japan (Inoue et al. 1998, Ueda and Kobayashi 2001), near the edge of a stand where adults concentrate because of their attraction to light (Igeta et al. 2003). Traps should be checked weekly.

Bait logs have also been proposed as a monitoring tool. Logs should be >1 m [~3 ¼ ft] long with a moisture content >60%; trap logs should be placed away from direct sunlight (Kobayashi and Ueda 2003, Kobayashi et al. 2004). Autoclaving logs extended their attractiveness (Ueda and Kobayashi 2004). This method cannot yet be recommended for use in the US because *Quercus crispula*, the species used for bait logs in Japan, is not widely available. Other tree species have not been tested.

Platypus quercivorus adults may be confused with morphologically similar wood-boring relatives. There are hundreds of species within the genus *Platypus* worldwide, though the majority are tropical (Schedl 1972, Barbosa and Wagner 1989, Farrell et al. 2001). Available keys to species in the US are incomplete (Chamberlin 1939, Wood 1979, Atkinson 2004). Seven species reportedly occur in the contiguous US, and four of these species occur in Florida (Wood 1979, Bright and Skidmore 2002, Atkinson 2004), where our analysis also predicts *P. quercivorus* might become established (Fig. 2): *P. quadridentatus* occurs in oaks and other hardwood species in the southern and southeastern US (Drooz 1985, Atkinson 2004). *P. flavicornis* is a secondary attacker of pines and occasionally in several hardwoods in the eastern and southern US (Drooz 1985, Atkinson 2004). *P. compositus* and *P. parallelus* reportedly reproduce in a wide variety of tree hosts including oaks, with the latter considered a particularly damaging species in the southeastern US and Mexico (Drooz 1985, Cibrián Tovar et al. 1995, Atkinson 2004, reviewed in CAB 2004). Adult beetles should be positively identified by a qualified taxonomist.

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Scolytus intricatus

Scientific Name

Scolytus intricatus Ratzeburg

Fig. 1. Adult *Scolytus intricatus.*
[Image from Ken Walker, www.invasive.org]

Synonyms:
Bostrichus
Coptogaster
Ekkoptogaster
Pygmaeoscolytus
Tubuloscolytus
Eccoptogaster intricatus
Eccoptogaster pygmaeus
Eccoptogaster picicolor
Scolytus carpini
Scolytus eccoptus
Scolytus penicillatus
Scolytus simmeli
Scolytus lenkoranus
Scolytus tiburtinus
Phthorophloeus spinulosus

Common Names

European oak bark beetle, oak bark beetle

Type of Pest

Bark, cambium and sapwood-boring beetle

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Coleoptera,
Family: Scolytidae, **Subfamily:** Scolytinae

Reason for inclusion in manual

CAPS Priority Pest (FY 2007)

Pest Description

“[Adult] Length 2.4-4.2 mm ... Stocky, broadly-oval, rusty-brown to dark-brown, rarely almost black. The elytra are of subdued shine, usually lighter than the rest of the body, the legs and pronotal anterior margin are rusty-bronze, the antennae and tarsi are light-rusty. ... The male frons on the level of the eyes is very slightly convex, nearly flat, with a transverse impression above the clypeus. The frons is covered with rather long, light-rusty hairs, particularly on the sides and above the clypeus, longitudinally, not very coarsely rugose and punctured on the sides. On either side above the base of the clypeus there is one characteristic narrow bunch of rusty setae, protruding towards the front” (Fig. 1) (Michalski 1973).

“The female frons is convex, impressed above the clypeus, longitudinally, not very coarsely rugose, scantly pubescent, the longest and densest pubescence being in the vicinity of the clypeus, above which bunches of rusty setae are lacking. The frons is broad in both sexes” (Fig. 2) (Michalski 1973).

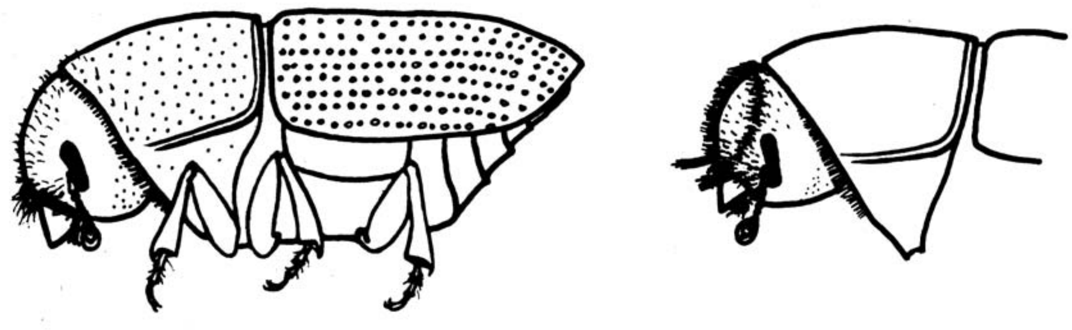

Fig. 2. Male (left) and female (right) *Scolytus intricatus*.
Note flatter frons and filiform setae above mandibles in male.
[Image reproduced from Grüne (1979)]

“Antennal Club. - Irregular-oval in shape, with two very indistinct, arcuate sutures and a faint triangular septum. The scape is nearly inconspicuous” (Michalski 1973).

“Pronotum. - The pronotum of both sexes is somewhat wider than long. shining, with the sides subparallel before the pronotal half length and strongly constricting anteriorly, with a distinct neck-like constriction. The punctation of the pronotum is well-defined, the punctures being small, longitudinal; punctures on the sides and in front are considerably larger, circular, partly conterminous. Sometimes the pronotum has a smooth line. The whole surface of the pronotum is micro-punctate. The scutellum is broad, triangular, with its apex raised, pubescent” (Michalski 1973).

“Elytra. - As wide as pronotum, or somewhat wider (in females) longer than that, with the sides subparallel before their half. The posterior angles of the elytra are with moderate arches, the elytral apices are rounded and slightly emarginate at the suture. The scutellar fovea is short. The striae and interstices are narrow, the punctures are markedly depressed, of similar size, the smaller ones being sometimes on the interstices, small, not very regularly spaced, particularly at the suture. The surface between punctures on the striae and interstices is obliquely corrugated, due to which the elytra seem to be dull (of subdued shine). The margin of the elytral apex is smooth, only the margins on the elytral posterior angles are slightly granular. On the interstices there are rows of short, erect, rusty hairs, longer at the apex and at the suture in its apical part” (Michalski 1973).

“Abdomen. - Obliquely ascendant in both sexes, without any projections, thickenings or tubercles, densely, finely punctured, covered densely with short and longer setae, the abdomen of the females being covered with coarser and more erect setae and setaceous hairs, particularly on the 5th sternite. The sides of the abdominal sternites in the females are with longer setaceous hairs. Between the setae there are recumbent, fine, short hairs, particularly on the 5th sternite" (Michalski 1973).

Duffy (1953) provides a useful key to genera of Scolytidae, and Grüne (1979) has a well illustrated key to species of *Scolytus*, including *S. intricatus*.

Biology and Ecology

Scolytus intricatus has 1-2 generations per year (Yates 1984a, reviewed in Haack 2001). Eggs, larvae, pupae, and adults generally occur in the cambial region of a host tree (Duffy 1953, Yates 1980, 1984a, reviewed in Haack 2001). Adults will leave bark to feed on young twigs (i.e., undertake maturation feeding) or find new hosts.

Adult are generally active between May and September, but the period of activity depends on temperature. Adults are unlikely to fly more than 100 m [330 ft] (reviewed in Yates 1984b, reviewed in Haack 2001). Newly emerged adults fly to host tree crowns and feed on tender young shoots and year-old growth for 2-3 weeks (Yates 1984b, reviewed in Haack 2001). Maturation feeding is not obligatory but may increase the fecundity of females (Yates 1980). Females comprise 50% of the population (Marković and Stojanović 2001). Mating typically occurs during maturation feeding or at the entrance of the egg gallery (Duffy 1953, Yates 1984a, reviewed in Haack 2001). Males or females can begin the construction of the egg gallery (reviewed in Haack 2001), but Yates (1980) suggests males are more likely to initially colonize a tree. Presumably males then release attractants to initiate a mass attack on a tree (Yates 1980), but an attractant for *S. intricatus* has not yet been identified. Hovorka et al. (2005) suggest the male-dominated colonization behavior is generally true for Nearctic, but not Palearctic, scolytids. Thus, the pattern described by Yates (1980) may not apply to *S. intricatus*. Adults seem to prefer weakened, dying trees or fallen branches (> 2 inches [5 cm] in diameter) for breeding, but smaller pieces of wood may be suitable. Egg galleries have been constructed in branches approximately ½-inch in diameter in laboratory studies (reviewed in Haack 2001). Rearing studies have demonstrated that adults will reproduce in fresh-cut logs (Yates 1984a, reviewed in Haack 2001). *Scolytus intricatus* is monogamous, and adults are not known to re-emerge from the host after mating (Yates 1984a, reviewed in Haack 2001).

Egg galleries are straight, single-armed, approximately 1-3 cm long [~½ -1¼ in], and oriented into wood; in some cases, the egg gallery superficially may penetrate the sapwood (Fig. 3) (Duffy 1953). A female will deposit 18-83 eggs in niches (1 egg/niche) on both sides of the gallery (Duffy 1953, Yates 1984a, reviewed in Haack 2001). Oviposition typically lasts 2 weeks. Eggs hatch in 10-14 days (Yates 1984a).

Fig. 3. Gallery produced by *Scolytus intricatus.*
[Image fromGyorgy Csoka, www.invasive.org]

Larvae develop through 5-6 instars (Yates 1984a, reviewed in Haack 2001). The number of larval instars may be difficult to determine for *S. intricatus* (Lekander 1968). Larvae overwinter between the third and fifth instars or as pupae (Yates 1984a, reviewed in Haack 2001). On average, overwintering larvae can withstand temperatures to -25.5°C [-14°F] but in some cases may survive as low as -29ºC [-20°F] (reviewed in Yates 1984a). Pupation occurs in the larval chamber in the late fall or from late spring to early summer either in the outer bark (measuring over 4 mm [~$^1/_8$ in] thick) or in the outer sapwood of hosts with thin bark (Yates 1984a, reviewed in Haack 2001). Survivorship of developing *S. intricatus* is quite high (32%) but depends on several factors including the ability of adults to find a suitable hosts and the presence of predator and parasitoids (Yates 1980, Marković and Stojanović 2001).

Scolytus intricatus is associated with several economically important fungi infecting hardwood tree species. In particular, this insect is a known vector for *Ophiostoma quercus* (reviewed in EPPO 1990, Leontovyč and Kunca 2000, Kirisits 2004) and may vector *Ceratocystis fagacearum* (Yates 1980). Both pathogens have severe negative affects on oak tree health. Frigimelica and Faccoli (1999) reported *S. intricatus* as a vector of the chestnut blight fungus, *Cryphonectria parasitica* in northeastern Italy. *Fusarium* spp., *Alternaria* spp., *Penicillium* spp., *Botyris* sp., *Gliocladum* sp., *Aspergillum* sp., *Sterilia*, and other fungi were isolated from *S. intricatus* adults that emerged from 50-60 year-old oak and chestnut trees infected with *C. parasitica* (Frigimelica and Faccoli 1999). Kubátová et al. (2004) isolated several species of *Geosmithia* and *Penicillium* from *S. intricatus*. *Geosmithia* spp. were found in all *S. intricatus* galleries and through all life cycle stages of this beetle (Kubátová et al. 2004).

Pest Importance

Scolytus intricatus is of moderate to high economic importance in Europe (Lieutier et al. 2004). It is generally considered a secondary pest which attacks trees that already dead or dying (reviewed in Marković and Stojanović 2003). In Hungary, approximately 547,000 ha [~1,300,000 acres] of broadleaf forest were infested by this insect; older oak trees were most severely affected (Lieutier et al. 2004). In Poland, *S. intricatus* is considered highly damaging. In portions of Europe, the insect is actively managed by forest sanitation that is achieved through selective thinning (reviewed in Lieutier et al. 2004). Chemical insecticides have also been applied to felled trees (reviewed in Lieutier et al. 2004).

The impact of the beetle on its own is difficult to measure. *Scolytus intricatus* is a vector of several Ascomycete fungi believed to be associated with vascular-wilt, oak-decline, and vascular-staining diseases in Europe (Édel'man and Malysheva 1959, Yates 1984a, reviewed in Haack 2001, Marković and Stojanović 2001, reviewed in CAB 2006). Establishment of *S. intricatus* would likely worsen the impact of oak wilt, caused by the already well established fungus, *C. fagacearum* (Juzwik 1999). See 'Biology and Ecology' for an expanded list of fungi associated with the insect.

Risks associated with *S. intricatus* have been evaluated previously. Haack (2001) considered the insect a very high risk to North American forests, but this assessment was uncertain. The potential for establishment, spread, economic harm, and environmental damage were rated 'high,' respectively. However, Haack (2001) noted that much of the environmental and economic damage would come from the fungi that the beetles may vector, not the beetles themselves. Perceived environmental damage from the beetles alone would be low (Haack 2001).

Symptoms

Trees infested by *S. intricatus* and its associated fungi may exhibit symptoms of general decline including sparse foliage and reduced growth. Infested trees may express signs of wilt, including yellow or red leaves on affected branches (reviewed in Édel'man and Malysheva 1959). Entrance and exit holes and boring dust from tunnel excavation may be observed. Adults may be observed on the bark surface of new hosts, at the entrance to newly constructed maternal galleries, or in the host tree crown maturation feeding on new shoot growth (up to 1 year old) (Yates 1984a, reviewed in Haack 2001).

Scolytus intricatus is the only species within the genus *Scolytus* in which the maternal gallery is transverse and non-branching (Duffy 1953, Yates 1984a). Larval tunnels reach an average length of 10 cm [ca. 4 in] and are at right angles to the maternal gallery (Yates 1984a).

Known Hosts

Scolytus intricatus reproduces and develops in several hardwood species, especially oak (Duffy 1953, Wood and Bright 1992). Hosts trees are typically weakened, declining or dead (Yates 1980, 1984a). *Castanea sativa*, *Corylus colurna*, *Quercus coccifera* and *Q. ilex* are preferred species in Serbia (Marković and Stojanović 2001). *Quercus* spp. are generally considered the preferred hosts (reviewed in Haack 2001).

Hosts	**References**
Aesculus sp. (buckeye)	(Bright and Skidmore 1997, CAB 2006)
Aesculus hippocastanum (horse chestnut)	(Michalski 1973, CAB 2006)
Alnus glutinosa (European alder)	(Bright and Skidmore 1997)

Hosts	**References**
Betula sp. (birch)	(Wood and Bright 1992)
Betula celtiberica (Iberian white birch)	(Bright and Skidmore 1997)
Betula pendula (common silver birch)	(Michalski 1973, CAB 2006)
Betula pubescens (downy birch)	(CAB 2006)
Betula verrucosa (white birch)	(Bright and Skidmore 1997, CAB 2006)
Carpinus sp. (hornbeam)	(Wood and Bright 1992, CAB 2006)
Carpinus betulus (European hornbeam)	(Michalski 1973, Bright and Skidmore 1997, CAB 2006)
Castanea sp. (chestnut)	(Duffy 1953)
Castanea sativa (European chestnut)	(Michalski 1973, Bright and Skidmore 1997, Marković and Stojanović 2001, CAB 2006)
Castanea vesca (sweet chestnut)	(Bright and Skidmore 1997)
Corylus sp. (hazelnut)	(Marković and Stojanović 2001, CAB 2006)
Fagus sp. (beech)	(Duffy 1953, Wood and Bright 1992, CAB 2006)
Fagus moesiaca	(Marković and Stojanović 2001)
Fagus orientalis (Oriental beech)	(Michalski 1973, Bright and Skidmore 1997)
Fagus sylvatica (common beech)	(Michalski 1973, Bright and Skidmore 1997, Kubátová et al. 2004, CAB 2006)
Ostrya sp. (hop-hornbeam)	(Wood and Bright 1992, CAB 2006)
Ostrya carpinifolia (European hop-hornbeam)	(Michalski 1973, Bright and Skidmore 1997, CAB 2006)
Parrotia persica (Persian parrotia)	(Michalski 1973, Bright and Skidmore 1997)
Populus sp. (poplar)	(Duffy 1953, Michalski 1973)
Populus alba (white poplar)	(Michalski 1973, Bright and Skidmore 1997)
Populus tremula (European aspen)	(Michalski 1973, Bright and Skidmore 1997)
Quercus sp. (oak)	(Duffy 1953, Yates 1984a, Heliövaara et al. 1991, Wood and Bright 1992, Frigimelica and Faccoli 1999, Haack 2001)
Quercus canariensis (Algerian oak)	(Bright and Skidmore 1997)
Quercus castaneaefolia (chestnut-leaf oak)	(Michalski 1973, Bright and Skidmore 1997)
Quercus cerris (European turkey oak)	(Michalski 1973, Bright and Skidmore 1997, Marković and Stojanović 2001, CAB 2006)

Hosts	**References**
Quercus coccifera (Kermes oak)	(Bright and Skidmore 1997, Marković and Stojanović 2001)
Quercus dalechampii	(Marković and Stojanović 2001, Kubátová et al. 2004, CAB 2006)
Quercus frainetto (Italian oak)	(Bright and Skidmore 1997, Marković and Stojanović 2001)
Quercus hartwissiana	(Michalski 1973)
Quercus ilex (holly oak)	(Bright and Skidmore 1997, Marković and Stojanović 2001)
Quercus lusitanica (Lusitanian oak)	(Michalski 1973, Bright and Skidmore 1997)
Quercus petraea (durmast oak)	(Michalski 1973, Bright and Skidmore 1997, Marković and Stojanović 2001, Kubátová et al. 2004, Hovorka et al. 2005, CAB 2006)
Quercus polycarpa	(Kubátová et al. 2004)
Quercus prinus var. *tomentosa*	(Michalski 1973)
Quercus pubescens (downy oak)	(Michalski 1973, Bright and Skidmore 1997)
Quercus pyrenaica (Pyrenean oak)	(Bright and Skidmore 1997)
Quercus robur (common oak)	(Michalski 1973, Bright and Skidmore 1997, Vrkočová et al. 1999, Marković and Stojanović 2001, Kubátová et al. 2004, Hovorka et al. 2005, CAB 2006)
Quercus rubra (northern red oak)	(Marković and Stojanović 2001)
Quercus virgiliana (Italian oak)	(Marković and Stojanović 2001)
Salix sp. (willow)	(Michalski 1973, Bright and Skidmore 1997, Vrkočová et al. 1999, CAB 2006)
Sorbus sp. (mountain ash)	(Bright and Skidmore 1997, CAB 2006)
Tilia cordata (small-leaf lime)	(CAB 2006)
Ulmus sp. (elm)	(Duffy 1953, Michalski 1973, Bright and Skidmore 1997, CAB 2006)
Ulmus carpinifolia (English elm)	(Michalski 1973)
Ulmus laevis (European white elm)	(Michalski 1973)
Zelkova carpinifolia (Caucasian zelkova)	(Michalski 1973, Bright and Skidmore 1997)

Known Distribution

Scolytus intricatus is reported from:

Location	**References**
Algeria	(Bright and Skidmore 1997)

Location	**References**
Austria	(Wood and Bright 1992, Haack 2001, CAB 2006)
Azerbaijan	(Michalski 1973, CAB 2006)
Belgium	(Wood and Bright 1992, Haack 2001, CAB 2006)
Bulgaria	(Wood and Bright 1992, CAB 2006)
Czech Republic	(Wood and Bright 1992, Vrkočová et al. 1999, Haack 2001, Kubátová et al. 2004, CAB 2006)
Denmark	(Heliövaara et al. 1991, Wood and Bright 1992, Haack 2001, CAB 2006)
Estonia	(Haack 2001, CAB 2006)
Finland	(Michalski 1973, Heliövaara et al. 1991, Wood and Bright 1992, Haack 2001, CAB 2006)
France	(Wood and Bright 1992, Haack 2001, CAB 2006)
Germany	(Wood and Bright 1992, Haack 2001, CAB 2006)
Greece	(Wood and Bright 1992, Haack 2001, CAB 2006)
Hungary	(Wood and Bright 1992, Haack 2001, CAB 2006)
Iran	(Bright and Skidmore 1997, Haack 2001, CAB 2006)
Italy	(Wood and Bright 1992, Haack 2001, CAB 2006)
Kazakhstan	(Michalski 1973)
Latvia	(Haack 2001, CAB 2006)
Luxembourg	(Wood and Bright 1992, Haack 2001, CAB 2006)
Morocco	(Michalski 1973, Wood and Bright 1992, Haack 2001, CAB 2006)
Netherlands	(Wood and Bright 1992, Vrkočová et al. 1999, Haack 2001, CAB 2006)
Norway	(Heliövaara et al. 1991, Wood and Bright 1992, Haack 2001, CAB 2006)
Poland	(Wood and Bright 1992, Haack 2001, CAB 2006)
Romania	(Wood and Bright 1992, Haack 2001, CAB 2006)
Russia	(Michalski 1973, Wood and Bright 1992, Haack 2001, CAB 2006)
Serbia	(Marković and Stojanović 2001)

Location	References
Slovakia	(Wood and Bright 1992, Haack 2001, Kubátová et al. 2004, CAB 2006)
Spain	(Wood and Bright 1992, Haack 2001, CAB 2006)
Sweden	(Michalski 1973, Heliövaara et al. 1991, Wood and Bright 1992, Haack 2001, CAB 2006)
Switzerland	(Wood and Bright 1992, Haack 2001, CAB 2006)
Tunisia	(Michalski 1973, Wood and Bright 1992, Haack 2001, CAB 2006)
Turkey	(Michalski 1973, Wood and Bright 1992, Haack 2001, CAB 2006)
United Kingdom	(Michalski 1973, Wood and Bright 1992, Haack 2001, CAB 2006)
Yugoslavia (former)	(Wood and Bright 1992, Haack 2001, CAB 2006)

Potential Distribution within the US

Based on the known distribution of *S. intricatus*, this insect is most likely to be associated with biomes characterized as temperate coniferous forest and temperate broadleaf-and-mixed forest. These biomes are common in the East, Pacific Northwest, and upper elevations of the Intermountain West. Together the biomes account for 47% of the area in the contiguous US.

A detailed biogeographic analysis of the distribution of *S. intricatus* in Scandinavia suggested the distribution of the species was most heavily influenced by precipitation and effective temperature summation (Heliövaara et al. 1991). A base temperature for the temperature summation was not reported. However, the probability of a *S. intricatus* being present generally was greater in areas with a higher effective temperature summation and decreased in areas with greater precipitation.

Survey

Pheromones for *S. intricatus* have not yet been identified, though Zhang et al. (2002) and Vrkočová et al. (1999) have searched for other potential semiochemicals from host and non-host plants. Hovorka et al. (2005) suggest that *S. intricatus* does not produce sex-specific aggregation pheromones, unlike many North American scolytids.

In Europe, the most common methods to monitor populations of *S. intricatus* include trap trees/logs, visual inspections, and questionnaires given to foresters or other natural resource managers (reviewed in Lieutier et al. 2004). Trap trees are simply felled trees that are left to be colonized by the insects. Trap trees should be approximately 3-4 inches [7-10 cm] in diameter and 13-16 ft [4-5 m] long (Édel'man and Malysheva 1959). Trap trees have been used in an effort to detect *S. intricatus* near the port city of Duluth, MN (S. Seybold, pers. comm.); no such beetles were found.

Scolytus intricatus could easily be confused with other *Scolytus* species. The genus *Scolytus* is comprised of approximately 55 species occurring in Asia, Europe and North America. Three closely related *Scolytus* species are indigenous to North America including the hickory bark beetle, *S. quadrispinosus*, and two fir engravers, *S. unispinosus* and *S. ventralis* (Furniss and Carolin 1977, Drooz 1985, reviewed in Haack 2001). Any suspected finds of *S. intricatus* should be confirmed by an appropriately trained entomologist.

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Arthropods
Moths

Adoxophyes orana[*]

Scientific Name
*Adoxophyes orana (*Fischer von Röslerstamm)

<u>Synonyms:</u>

Adoxophyes orana Bradley, 1952
Adoxophyes reticulana Chambon & d'Aguilar, 1974
Adoxophyes reticulana Hübner
Capua reticulana Hübner
Cacoecia reticulana
Capua orana
Tortrix orana Fischer von Röslerstamm
Tortrix reticulana
Capua congruana
Adoxophyes tripsiana
Adoxophyes fasciata Walsh
Acleris reticulana
Adoxophyes congruana Walker
Tortrix orana Fischer von Röslerstamm, 1834
Tortrix reticulana Hübner, 1818

Fig. 1. Adult *Adoxophyes orana* in repose.
[Image from Hania Arentsen, #UGA2102086, www.invasive.org]

The species *Adoxophyes fasciata* is technically a subspecies of *A. orana*. "The subspecies has so far been generally known from continental Europe" (Yasuda 1998). *Adoxophyes fasciata* is considered a synonym of *A. orana*. *Adoxophyes orana fasciata* Walsingham has the following synonyms:

Adoxophyes fasciata Walsingham, 1900
Adoxophyes orana fasciata
Adoxophyes orana

Common Names
summer fruit tortrix
reticulated tortrix

Type of Pest
Moth, leafroller, defoliator

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[*] This document was largely excerpted from the report: Davis, E.E., S. French, and R.C. Venette. 2005. Mini-Risk Assessment: Summer Fruit Tortrix Moth, *Adoxophyes orana* (Fisher von Röslerstamm, 1834) [Lepidoptera: Tortricidae]. Available on-line at www.aphis.usda.gov/ppq/ep/pestdetection/pra/aoranapra.pdf

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for inclusion in manual

CAPS Priority Pest (FY 2007)

Pest Description

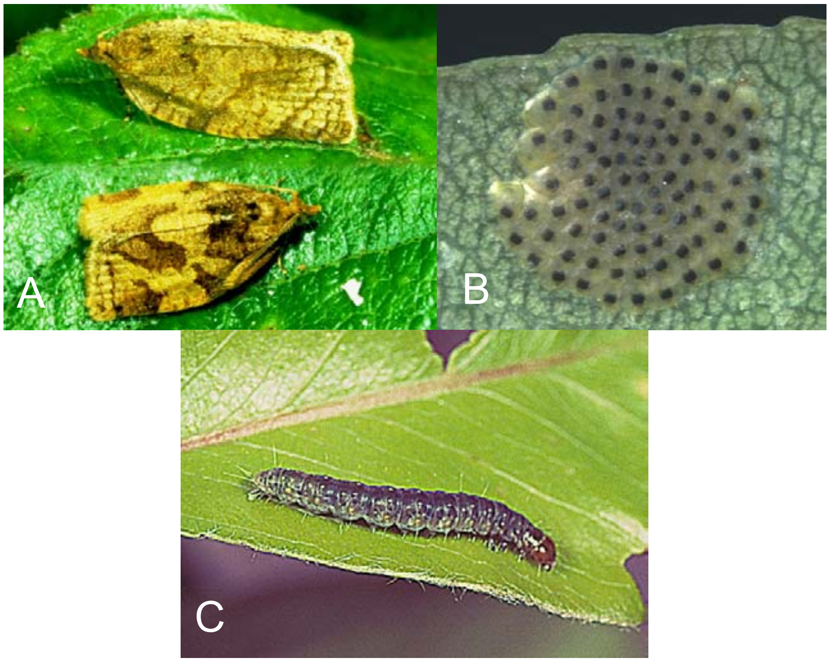

Fig. 2. *Adoxophyes orana*: (A) adult female [above] and male [below]; (B) egg "raft" showing black head capsules of larvae prior to hatch on *Malus* leaf; and (C) larva on *Pyrus* sp.
[Images by R. Coutin/OPIE (INRA 2005)].

"[Forewing length]. Male 10.0-11.0 mm, Female 11.0-13.0 mm. The forewing of the female is rather dull greyish brown, while in the male the coloration is brighter and is a yellowish brown. The male has a fold that extends about ½ of the length of the costa, and the fold is lined with whitish small glandular scales" (Yasuda 1998).

"[Wingspan] Male 15-19 mm, Female 18-22 mm. Sexual dimorphism pronounced; antenna of male shortly ciliate, forewing with broad costal fold from base to about one-third, markings usually conspicuous, contrasting with paler ground colour; female usually larger, antenna minutely ciliate, forewing without costal fold, with darker general coloration and less contrasting markings" (Bradley et al. 1973).

Male: "Ground colour of forewing light greyish brown; markings dark brown suffused with ochreous; outer margin of basal fasciae poorly defined, oblique to middle; median fascia narrow, margins irregular, usually constricted at middle before emitting strong tornal spur; pre-apical spot broken and reduced, emitting a strong stria extending to the tornal area, and a second much thinner stria parallel with termen. Hindwing grey" (Bradley et al. 1973).

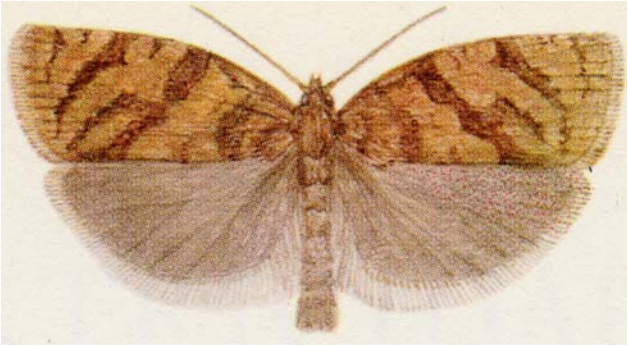

Fig. 3. Male *A. orana.*
[Color plate reproduced from Bradley (1973).]

Female: "Forewing ground colour greyish brown; markings essentially as in male but more subdued and often partially obsolete. Hindwing grey" (Bradley et al. 1973).

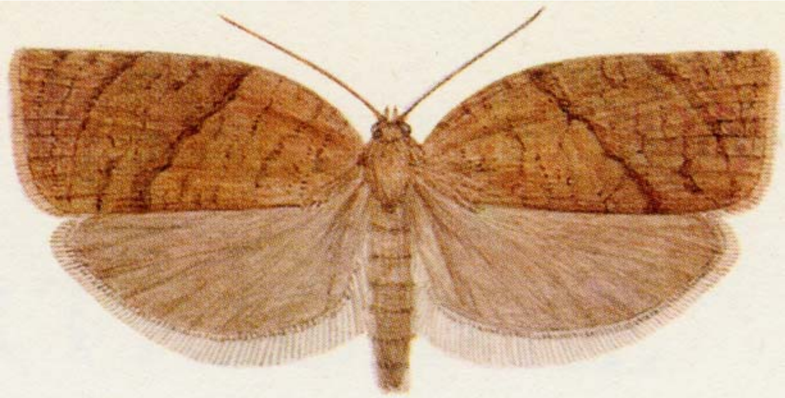

Fig. 4. Female *A. orana.* The larger image size reflects the larger relative size of females.
[Color plate reproduced from Bradley (1973).]

Biology and Ecology

In the Netherlands and much of Europe, *A. orana* has two generations annually. First generation larvae feed in summer (typically June or July) on leaves, buds, flowers and developing fruit. Larvae from the second generation feed in fall in preparation for diapause as a second or third instar (Fluckiger and Benz 1982). The insect overwinters as a diapausing larva. A partial third generation is possible if warm temperatures persist in fall; however if larvae do not successfully enter diapause, they are likely to die (de Jong et al. 1971, Barel 1973, de Jong and Van Dieren 1974, Berlinger and Ankersmit 1976, Stamenkovic and Stamenkovic 1984, Charmillot and Brunner 1989, 1990, Stamenkovic et al. 1999). Feeding resumes in the spring, typically April, on young leaves, buds and flowers (Fluckiger and Benz 1982).

In northwestern Europe, adults are present from late May to late June (first generation), late July to early September (second generation), and October (third generation) (CAB 2004). Flight periods of the first and second generation may overlap (Whittle 1985). Flight lasts approximately 4 weeks (Barel 1973). Moths fly at temperatures above 13°C [55°F] (Whittle 1985). Males precede females in flight by a few days and may disperse up to 1,300 ft [400 m]. Female dispersal is limited (Barel 1973, CAB 2004). Mating occurs at night or in early morning hours, about a day after emergence (de Jong et al. 1971, Whittle 1985, He et al. 1996). Adults rest on leaves within the tree canopy during the day and become active at dusk (Bradley et al. 1973).

A single female can lay 200-400 eggs (Bradley et al. 1973, Whittle 1985, CAB 2004). Eggs are laid on upper and lower leaf surfaces in masses of 4-150 or more (CAB 2004). Eggs will occasionally be deposited on fruit (Bradley et al. 1973) or on tree trunks if population density is high (CAB 2004). Egg production depends on temperature. More eggs are produced as temperatures rise above 15°C [59°F]; fewer eggs are produced below 13°C [55°F]; and no eggs are produced at or below 9°C [48°C] (de Jong et al. 1971, Charmillot et al. 1984, Whittle 1985, CAB 2004). Eggs require about 90 degree days [°C] above 10°C [50°F] to hatch (Charmillot and Megevand 1983), which equates to about 8-20 days in the field, depending on temperature (Bradley et al. 1973, de Jong 1980, CAB 2004).

The insect develops through 5-6 instars (CAB 2004). Overwintering larvae begin feeding in the spring after 67 degree days [°C] accumulate above a threshold of 9-10°C [48-50°F] (Charmillot and Megevand 1983, Whittle 1985). In preparation for feeding, larvae will create protected feeding sites by binding leaves, flower buds or other plant parts together with silk (CAB 2004). Late instars may be found in the crown on new shoot growth (CAB 2004). Summer generation larvae complete development on average in 430 degree days [°C] above a threshold of 7-8°C [44-46°F] (Charmillot and Megevand 1983, Whittle 1985).

Diapause is induced in larvae by short day length,<12-16 h of light at 20-25°C [68-77°F] (Barel 1973, Berlinger and Ankersmit 1976, Bonnemaison 1977, Whittle 1985). Length of diapause influences when diapause will be terminated (Milonas and Savopoulou-Soultani 2004). Larvae pupate in leaves bound together with silk or within a silk cocoon in leaves, on twigs, in old mummified fruit, or trunks (Bradley et al. 1973, CAB 2004). Pupation requires an accumulated 90 degree days [°C] above a threshold of 10°C [50°F] (Charmillot and Megevand 1983).

Pest Importance

Adoxophyes orana has the potential to feed on oak and other forest species (CAB 2004). However, the insect is better known as a major pest of fruit crops, particularly apple and pear, in temperate regions (Whittle 1985, Hill 1987, INRA 2005).

Establishment of *A. orana* in the US may have limited adverse environmental consequences. Although *A. orana* will feed on foliage and young shoots (CAB 2004), this feeding may not significantly affect plant growth (INRA 2005). The impact of *A. orana* on forest productivity has not been well studied.

The economic impact of *A. orana* is difficult to measure because it frequently occurs in mixed populations with other closely related species, and damage can result from the activity of secondary pests (Whittle 1985). Feeding directly on fruit can cause tremendous reductions in the quantity and quality of fruit. Crop losses from 10-50% have been attributed to this insect in fruit growing regions. In the Netherlands, damage in 33,000 ha of apples amounted to $1.2 million in the late 1980s (de Jong et al. 1971, Whittle 1985). External feeding may also enable the attack of secondary organisms which further damage the crop and reduce shelf life (de Jong et al. 1971, Whittle 1985, INRA 2005).

Risks associated with *A. orana* have been evaluated previously, at least in part. For example, when evaluating the potential importation of Asian pear, Australia judged the overall risk potential to be high (BA-AQIS 2003). Cave and Lightfield (1997) recognized that *A. orana* might attack fragrant and ya pear in China but did not consider it likely that the pest would remain associated with the fruit if it were allowed to be shipped to the US.

Symptoms

External feeding will be visible on leaves and fresh growth of twigs. Feeding will deform leaves and create areas with dead tissue. Leaves may appear wilted, yellow, shredded, or dead. Leaves are likely to be rolled or folded and held together with silk webbing. Feeding on new growth of twigs will leave lesions. If the insect is feeding in flowers, external feeding damage and silk webbing will be evident. In all areas where the insect has fed, frass should also be visible. (ECOPORT 2000).

Summer-generation larvae feed extensively and severely damage fruit (de Jong and Beeke 1976). Feeding on fruits or pods causes scabs or pitting on the fruit and fruit deformation (skin or general shape); silk webbing and frass may be present (ECOPORT 2000). On fruit crops, larvae prefer to feed sheltered under a leaf bound to fruit with silk (Bradley et al. 1973)

Known Hosts

Adoxophyes orana is not host specific. It reportedly feeds and develops on more than 50 plant species in multiple families (Table 1). Although the host range includes several forest species, *A. orana* may feed preferentially on apples, pears and other rosaceous hosts (reviewed in INRA 2005).

Hosts	**References**
Acer sp. (maple)	(de Jong et al. 1971)

Hosts	References
Acer campestre (maple, common)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Alnus sp. (alder)	(Janssen 1958, Balachowsky 1966, de Jong et al. 1971, Barel 1973, Whittle 1985, CAB 2004)
Betula sp. (birch)	(Janssen 1958, Balachowsky 1966, de Jong et al. 1971, Barel 1973, de Jong and Minks 1981, Savopoulou-Soultani et al. 1985, Whittle 1985, CAB 2004)
Camellia sp. (tea[2])	(de Jong et al. 1971, Barel 1973, CIE 1982)
Camellia sinensis (tea[2])	(Whittle 1985)
Carpinus sp. (hornbeam)	(de Jong et al. 1971)
Carpinus betulus (hornbeam, European)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Chenopodium album (lambsquarters)	(Barel 1973)
Convolvulus arvensis (field bindweed)	(Barel 1973)
Corylus sp. (filbert)	(Whittle 1985)
Cotoneaster dielsiana (cotoneaster)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985)
Crataegus sp. (hawthorn)	(Janssen 1958, Balachowsky 1966, de Jong et al. 1971, Barel 1973, Van Der Pers 1981, Savopoulou-Soultani et al. 1985, Whittle 1985, CAB 2004)
Cydonia oblonga (quince)	(Janssen 1958, Balachowsky 1966, de Jong et al. 1971, Barel 1973, Savopoulou-Soultani et al. 1985, Whittle 1985, Stamenkovic et al. 1999, CAB 2004)
Damasonium sp. (damson)	(de Jong et al. 1971)
Fagus sylvatica (common beech)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Forsythia suspensa (forsythia, weeping)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Fragaria sp. (strawberry)	(de Jong et al. 1971, Whittle 1985)
Fraxinus sp. (ash)	(de Jong et al. 1971, Whittle 1985)
Gossypium sp. (cotton)	(de Jong et al. 1971, Savopoulou-Soultani et al. 1985, Shu et al. 2002)
Gossypium herbaceum (cotton, Arabian)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Gossypium hirsutum (cotton, upland)	(Whittle 1985)
Humulus sp. (hop)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)

Hosts	**References**
Laburnum sp. (laburnum)	(Balachowsky 1966, de Jong et al. 1971, Barel 1973, Whittle 1985)
Laburnum anagyroides (golden chain tree)	(Janssen 1958, CAB 2004)
Ligustrum sp. (privet)	(Janssen 1958, Balachowsky 1966, de Jong et al. 1971, Barel 1973, Savopoulou-Soultani et al. 1985, Whittle 1985, CAB 2004)
Lonicera sp. (honeysuckle)	(de Jong et al. 1971, Savopoulou-Soultani et al. 1985)
Lonicera caprifolium (Italian woodbine)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985)
Lonicera xylosteum (honeysuckle, fly)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Malus baccata, M. baccata jackii (crabapple, Siberian)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Malus domestica (apple)	(Barel 1973, reviewed in CIE 1982, Zhang et al. 1990)
Malus pumila (apple, paradise)	(Janssen 1958, Balachowsky 1966, Barel 1973, CAB 2004)
Malus sylvestris (crabapple, European)	(Whittle 1985)
Medicago sp. (alfafla)	(Janssen 1958, Balachowsky 1966, Barel 1973, Savopoulou-Soultani et al. 1985, Whittle 1985, CAB 2004)
Menyanthes trifoliata (buckbean)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985)
Morus sp. (mulberry)	(Im and Paik 1982)
Parrotia sp. (ironwood)	(Janssen 1958, Balachowsky 1966, Barel 1973, Savopoulou-Soultani et al. 1985, Whittle 1985)
Physalis peruviana (Peruvian groundcherry)	(Whittle 1985)
pillar tree (scientific name unknown)	(Hassan and Rost 1993)
Pistacia lentiscus (mastic tree)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Pistacia sp. (pistachio)	(Savopoulou-Soultani et al. 1985)
Populus sp. (poplar)	(Janssen 1958, Balachowsky 1966, de Jong et al. 1971, Barel 1973, Savopoulou-Soultani et al. 1985, Whittle 1985, CAB 2004)
Potentilla sp. (cinquefoil)	(Barel 1973)
Prunus sp.	(Van Der Pers 1981, Savopoulou-Soultani et al. 1985)

Hosts	**References**
Prunus armeniaca (apricot)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, Stamenkovic et al. 1999, CAB 2004)
Prunus avium (cherry, sweet)	(Janssen 1958, Balachowsky 1966, Barel 1973, CIE 1982, CAB 2004)
Prunus cerasus (cherry, sour)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, Stamenkovic et al. 1999)
Prunus sp. (plum)	(de Jong et al. 1971, de Jong and Minks 1981, Stamenkovic et al. 1999, Hrudova 2003)
Prunus domestica (plum, European)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Prunus domestica subsp. *insititia*, *Prunus domestica* subsp. *syriaca* (=*P. insistitia syriaca*)	(Janssen 1958, Balachowsky 1966, Barel 1973)
Prunus insistitia	(Balachowsky 1966, Whittle 1985)
Prunus padus (cherry, bird)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Prunus persica (peach leaves[1])	(Milonas and Savopoulou-Soultani 2000)
Prunus persica (peach)	(Janssen 1958, Balachowsky 1966, Barel 1973, CAB 2004)
Prunus triloba (almond tree, flowering)	(Janssen 1958, Balachowsky 1966, Barel 1973, Whittle 1985, CAB 2004)
Pyrus sp.	(Savopoulou-Soultani et al. 1985)
Pyrus communis (pear leaves[1])	(Fluckiger and Benz 1982)
Pyrus communis (pear)	(Balachowsky 1966, Honma 1970, de Jong et al. 1971, CIE 1982)
Pyrus communis (pear, European)	(Janssen 1958, Barel 1973, Whittle 1985, CAB 2004)
Pyrus pyrifolia (pear, Asian 'Shandong'), *P. ussuriensis* var. *viridis* (pear, Asian 'Ya') fruit, leaves and shoot	(BA-AQIS 2003)
Quercus sp. (oak)	(Janssen 1958, Barel 1973, Savopoulou-Soultani et al. 1985, Whittle 1985)
Quercus robur (oak, English[1])	(Fluckiger and Benz 1982)
Rhododendron catawbiense (Catawba rosebay[1])	(Fluckiger and Benz 1982)
Ribes sp.	(de Jong et al. 1971, Savopoulou-Soultani et al. 1985, Whittle 1985)

Hosts	References
Ribes nigrum (currant, black)	(Janssen 1958, de Jong et al. 1971, Barel 1973, Whittle 1985, CAB 2004)
Ribes rubrum (currant, red)	(Janssen 1958, de Jong et al. 1971, Whittle 1985, CAB 2004)
Ribes uva-crispa var. *sativum* (=*R. grossularia*) (gooseberry, European)	(Janssen 1958, Barel 1973, Whittle 1985, CAB 2004)
Rosa sp. (roses)	(Janssen 1958, de Jong et al. 1971, Barel 1973, de Jong and Minks 1981, Savopoulou-Soultani et al. 1985, CAB 2004)
Rosa canina (dog rose)	(Janssen 1958, Barel 1973, Whittle 1985, CAB 2004)
Rubus sp.	(de Jong et al. 1971, Savopoulou-Soultani et al. 1985)
Rubus fruticosus (blackberry, shrubby)	(Janssen 1958, Barel 1973, Whittle 1985, CAB 2004)
Rubus idaeus (raspberry)	(Janssen 1958, de Jong et al. 1971, Barel 1973, Whittle 1985, CAB 2004)
Rumex sp. (dock)	(Barel 1973)
Rumex obtusifolius (dock, bitter[1])	(Fluckiger and Benz 1982)
Salix sp. (willow)	(de Jong et al. 1971, Van Der Pers 1981, Savopoulou-Soultani et al. 1985)
Salix caprea (willow, goat)	(Janssen 1958, Barel 1973, Whittle 1985, CAB 2004)
Salix viminalis (willow, basket)	(Janssen 1958, Barel 1973, Whittle 1985, CAB 2004)
Solanum sp. (nightshade)	(Savopoulou-Soultani et al. 1985)
Solanum dulcamara (nightshade, climbing)	(Janssen 1958, Barel 1973, Whittle 1985)
stone fruit	(Sziraki 1984)
Symphoricarpos albus (common snowberry)	(Whittle 1985, CAB 2004)
Symphoricarpos albus var. *albus* (=*S. racemosus*) (common snowberry)	(Janssen 1958, Barel 1973)
Syringa sp. (lilac)	(de Jong et al. 1971, CIE 1982, Savopoulou-Soultani et al. 1985)
Syringa vulgaris (lilac, common)	(Janssen 1958, Barel 1973, Whittle 1985, CAB 2004)
Tilia sp. (basswood)	(Janssen 1958, de Jong et al. 1971, Barel 1973, Savopoulou-Soultani et al. 1985, Whittle 1985, CAB 2004)

Hosts	References
Ulmus sp. (elm)	(de Jong et al. 1971, Savopoulou-Soultani et al. 1985, Whittle 1985)
Ulmus campestris (elm, English or Wych)	(Janssen 1958, Barel 1973)
Ulmus minor (elm, European field)	(CAB 2004)
Urtica sp. (nettle)	(Janssen 1958, Barel 1973, Whittle 1985)
Urtica dioica (nettle, stinging)	(de Jong et al. 1971)
Vaccinium sp. (blueberry)	(Janssen 1958, Barel 1973, Savopoulou-Soultani et al. 1985, Whittle 1985, CAB 2004)
Vicia faba (horsebean)	(Barel 1973)
Vitis vinifera (grapevine)	(Savopoulou-Soultani et al. 1985)

1. Laboratory experiments.
2. Tea may or may not be a host of *A. orana*, possibly a misidentification due to taxonomic confusion. Tea was reported as a host associated with *A. orana* "tea form" which was later named *A. honmai,* (Yasuda 1998). However according to Barel (1973), the “tea strain” was likely a synonym of *A. orana*.

Known Distribution

Adoxophyes orana occurs throughout much of Europe and Asia and is not known to occur in the United States (CIE 1982). Much of its range may be adventive. The insect has successfully invaded England (Cross 1996) and Greece (Savopoulou-Soultani et al. 1985).

Location	References
Albania	(Kapidani and Duraj 1991)
Armenia	(CAB 2004)
Austria	(Barel 1973, CIE 1982, Whittle 1985, CAB 2004)
Azerbaijan	(CAB 2004)
Belgium	(de Jong et al. 1971, Barel 1973, CIE 1982, Vanwetswinkel and Soenen 1983, Verheyden 1984, Whittle 1985, Charmillot and Brunner 1989, CAB 2004)
Bosnia and Herzegovina (Belgrade)	(Savopoulou-Soultani et al. 1985, Stamenkovic et al. 1999)
Bulgaria	(CIE 1982, Whittle 1985, CAB 2004)
China	(Meng et al. 1978, de Jong and Minks 1981, CIE 1982, Ankersmit 1985, Whittle 1985, Zhang et al. 1990, Cave and Lightfield 1997, Zhou et al. 1997, Shu et al. 2002, BA-AQIS 2003, Ding et al. 2003, CAB 2004)

Location	References
Czechloslovakia (former), Czech Republic	(Barel 1973, Hrudova 2003)
Denmark	(Barel 1973, CIE 1982, Whittle 1985, CAB 2004)
England	(de Jong et al. 1971, Barel 1973, Alford 1979, Alford et al. 1979, CIE 1982, Fluckiger and Benz 1982, Langmaid 1984, Whittle 1985, Baker 1991, Morgan 1992, Morgan and Solomon 1993, Cross 1996, Solomon and Morgan 1996, Jay and Cross 1998, Spence 1998, Cross et al. 1999b, Stamenkovic et al. 1999, CAB 2004)
Finland	(Barel 1973, CIE 1982, Whittle 1985, CAB 2004)
France	(Barel 1973, CIE 1982, Whittle 1985, Gendrier 1988, Charmillot and Brunner 1989, CAB 2004)
Georgia, Republic	(CAB 2004)
Germany	(de Jong et al. 1971, Barel 1973, de Jong and Minks 1981, CIE 1982, Dickler 1982, 1984, Whittle 1985, Charmillot and Brunner 1989, Hassan and Rost 1993, Kienzle et al. 1997a, Kienzle et al. 1997b , Cross et al. 1999a, Potting et al. 1999, CAB 2004)
Greece (Naoussa area)	(Savopoulou-Soultani et al. 1985, Charmillot and Brunner 1989, Milonas and Savopoulou-Soultani 1999, 2000, 2004)
Holland	(Barel 1973, Stamenkovic et al. 1999)
Hungary	(Barel 1973, CIE 1982, Sziraki 1984, Whittle 1985, Balazs 1992, 1997, Balazs et al. 1997, CAB 2004)
Italy	(de Jong et al. 1971, Barel 1973, de Jong and Minks 1981, CIE 1982, Whittle 1985, Charmillot and Brunner 1989, Rama et al. 1997, Stamenkovic et al. 1999, CAB 2004)

Location	References
Japan[1]	(Honma 1970, de Jong et al. 1971, Minks et al. 1971, Honma 1972, Barel 1973, Tamaki et al. 1976, de Jong and Minks 1981, CIE 1982, Im and Paik 1982, Ankersmit 1985, Whittle 1985, CAB 2004)
Korea	(Im and Paik 1982, Goh et al. 1984, Lee et al. 1992, Jo and Kim 2001, CAB 2004)
Netherlands	(Minks 1969, de Jong et al. 1971, Minks and Noordink 1971, Minks et al. 1971, Barel 1973, Minks and Voerman 1973, Minks and de Jong 1975, Berlinger and Ankersmit 1976, de Jong and Beeke 1976, Ankersmit 1980, de Jong 1980, de Jong and Minks 1981, CIE 1982, Fluckiger and Benz 1982, Van Der Kraan and van Deventer 1982, Vanwetswinkel and Soenen 1983, Ankersmit 1985, Whittle 1985, de Reede and de Wilde 1986, Blommers et al. 1987, Helsen and Blommers 1989, Malais and Ravensberg 1993, Neumann et al. 1993, Minks et al. 1995, Jay and Cross 1998, CAB 2004, Milonas and Savopoulou-Soultani 2004)
Norway	(Barel 1973, CIE 1982, Whittle 1985, CAB 2004)
Poland	(Barel 1973, CIE 1982, Whittle 1985, CAB 2004)
Romania	(Barel 1973, CIE 1982, Whittle 1985, Ghizdavu 1986, CAB 2004)
Russia	(de Jong et al. 1971, Barel 1973, de Jong and Minks 1981, CIE 1982, Ankersmit 1985, Whittle 1985, Mottus et al. 2001, CAB 2004)
Scandinavia (except northern regions)	(de Jong et al. 1971)
Serbia and Montenegro	(CIE 1982, Injac and Dulic 1982, Stamenkovic and Stamenkovic 1985, Stamenkovic 1988, Krnjajic et al. 1993, Stamenkovic et al. 1999, CAB 2004)
Spain	(Barel 1973, CIE 1982, Whittle 1985, CAB 2004)

Location	References
Sweden	(Barel 1973, CIE 1982, Whittle 1985, CAB 2004)
Switzerland	(Barel 1973, de Jong and Minks 1981, CIE 1982, Fluckiger and Benz 1982, Baumgaertner and Charmillot 1983, Charmillot et al. 1984, Whittle 1985, Baumgaertner et al. 1988, Sechser and Engelhardt 1988, Charmillot and Brunner 1989, Minks et al. 1995, CAB 2004)
Ukraine	(Whittle 1985, CAB 2004)
Yugoslavia	(de Jong and Minks 1981, Whittle 1985, Charmillot and Brunner 1989, CAB 2004)

1. In Japan, *A. orana* "tea form" was later named as one of two new species, *A. honmai* or *A. dubia*, all of which occur in Japan and are not easily distinguishable. Possible misidentification.

Potential Distribution within the US

The currently reported distribution of *A. orana* suggests that the pest may be most closely associated with biomes characterized as tropical and subtropical moist, broadleaf forests, and temperate, broadleaf and mixed forests. Consequently, we estimate that approximately 29% of the continental US would have a climate suitable for establishment by *A. orana*. Known hosts, especially cultivated Rosaceae (e.g., apple, pear, apricot, and peach) and non-cultivated hardwoods (e.g., ash, alder, birch, cottonwood, and elm), are common in these climatically suitable areas.

Survey

Several monitoring techniques have been developed and applied to *A. orana*. The most effective approach involves sex-pheromone-baited traps. El-Sayed (2004) and Witzgall et al. (2004) summarize semiochemicals that have been identified for *A. orana*. The sex pheromone is a blend of (*Z*)-9-tetradecenyl acetate and (*Z*)-11-tetradecenyl acetate (Tamaki et al. 1971, Meijer et al. 1972). These two compounds are most attractive to males in a 9:1 blend of (Z)-9:(Z)-11 isomers; *E*-isomers of either compound had a strong inhibitory effect (Minks and Voerman 1973). CAB (2004) suggests that an 80:20 mixture of (Z)-9:(Z)-11 tetradecenyl acetate is most attractive, but this statement is incorrect. The 9:1 pheromone blend is available commercially as Adoxomone (Murphy Pherocon^{TM} Summer Fruit Tortrix Moth Attractant) for use with Pherocon 1C traps [Zoecon Corp] (Barel 1973, Alford 1979). Polyethylene caps treated with 100 µg of the pheromone blend remain attractive for >7 weeks (Minks and Voerman 1973), but baits should be changed about every 6 weeks (Alford 1979).

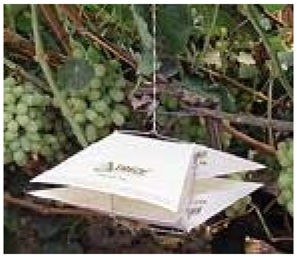

Fig. 5. Pherocon 1C trap [Image from Trécé Corp.].
Mention of a product name does not constitute endorsement.

The attractiveness of a trap extends more than 10 m [ca 33 ft] (Shirasaki 1989). For intensive monitoring within orchards, traps should be placed 15-20 m [ca. 50-65 ft] apart (Alford 1979, Hrudova 2003). For general monitoring and surveys, van der Kraan and van Deventer (1982) recommend 45 m [ca. 150 ft] between traps. Traps should be placed approximately 1.5 m [5 ft] above the ground (Barel 1973, Minks and de Jong 1975, Hrudova 2003); traps at other heights capture substantially fewer moths (Shirasaki 1989). When traps are deployed, night temperatures should be >14°C [57°F], the temperature threshold for adult flight (Barel 1973).

Dickler (1982) effectively used pheromone baited traps for a regional survey for *A. orana* in East Germany, as did Goh et al. (1984) in Suweon, North Korean. With a high diffusion rate of 15.2 mg/(ha·h), (Charmillot 1981) used the 9:1 blend to effectively disrupt mating; a diffusion rate of 7.3 mg/(ha·h) was ineffective. Similar results were obtained by Neumann et al. (1993). There is not a strong relationship between trap capture and plant damage (Alford et al. 1979).

Occasionally, pheromone traps will attract non-target species. Adoxomone also attracted the noctuid moth *Ceramica pisi* (Alford 1979). However, Hrudova (2003) failed to collect any non-target moths in traps baited for *A. orana*. Hrduova (2003) did note that *A. orana* was attracted very infrequently to traps with semiochemicals for *Cydia molesta*.

As an alternative to pheromone traps, Robinson light traps (Alford 1979) with 125W mercury vapor bulbs, 125W black light bulb, or 100W flood light can be used (Barel 1973). While sex pheromone traps attract males of a targeted species, light traps non-selectively draw in many flying insects. Minks (1969) captured more specimens of *A. orana* during the first generation in a trap baited with virgin females than in a light trap but obtained the exact opposite result during the second generation. Alford et al. (1979) also reported ca. 30% more moths in light traps than in pheromone traps; however if counts were corrected for the sex bias, pheromone traps captured more specimens than light traps (Alford 1979).

Visual sampling and beat sampling may also be used to inspect trees for eggs and larvae. Both methods are time consuming; 100 shoots should be processed using the beat method (de Jong 1980). For visual surveys, Pralja et al. (1992)

recommend the trunk and "all first order skeleton branches" at 1 m [3.3 ft] from the trunk to sample for eggs and "1.5 m [5 ft] long peripheral parts of four skeleton branches (one branch at each tree side) of the second order (for caterpillars)." Larvae tend to be aggregated among trees (Qiu et al. 1999). Visual sampling or beat sampling are not commonly recommended.

Adoxophyes orana may occur in mixed populations with closely related or morphologically similar species. By their very secretive nature, leafrollers are difficult to detect. Distinguishing between males and females of adult *Adoxophyes* is difficult in general (Balachowsky 1966). According to Yasuda (1998), "The extensive color and pattern variation of the forewing and morphological resemblance among *Adoxophyes* species have created difficulties in the identification of the species." Any identification should be confirmed by an appropriately trained entomologist.

References

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Archips xylosteanus

Scientific Name
Archips xylosteanus (Linnaeus)

<u>Synonyms:</u>
Archips xylosteana (Linnaeus)
Cacoecia xylosteana
var. *pallens* Kennel
Phalaena Tortrix xylosteana Linnaeus
Phalaena Tortrix desana Villers
Pyralis hybernana Fabricius
Pyralis obliquana Fabricius
Tortrix characterana Hübner
Tortrix westriana Thunberg

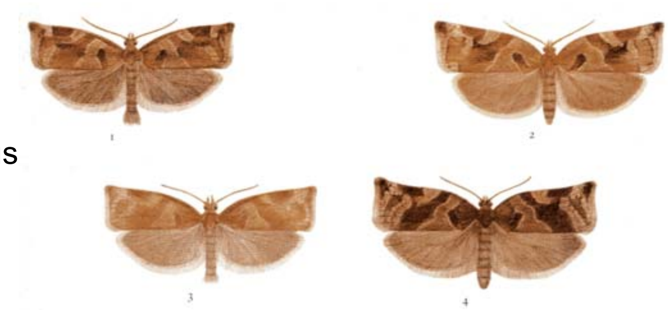

Fig. 1. Illustrations of adult *Archips xylosteanus* with males on left and females on right
[Image reproduced from Bradley et al. (1973)]

Common Names
variegated golden tortrix
apple leafroller
brown oak tortrix
twist moth
forked red barred moth

Type of Pest
moth, leafroller, defoliator

Taxonomic Position
Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for inclusion in manual
Recent detection in North America & classification as high risk (Spears 2006)

Pest Description
Adult: No clear sexual dimorphism (Bradley et al. 1973, Toimil 1987). Variation in coloration and forewing markings; "forewings whitish ochreous with ochreous brown or reddish brown, pale edged markings; a black-brown dot at disc; subterminal marking pistol-shaped in males. Hindwings greyish-brown" (Meijerman and Ulenberg 2000).

Male: wingspan 15-21 mm, "Forewing ground colour whitish ochreous, partially suffused with olive-grey; markings reddish brown, thinly edged with clear ground colour; inner margin of median fascia sinuate, pre-apical spot semi-ovate, usually contiguous with stria-like marking to tornus" (Bradley et al. 1973).

Fig. 2. Adult *Archips xylosteanus*
[Image from: http://nrm.museum/en/svenska_fjarilar/a/archips_xylosteana.html]

Female: wingspan 16-23 mm, "Forewing ground colour as in male; markings less reddish, often darker. Hindwing grey, apical area sometimes tinged with yellow or cupreous" (Bradley et al. 1973).

"Egg: Cylindrical, greenish eggs are deposited in oval masses which are variable in size (3x7 to 4.5x10 mm). Eggs are covered with a brown secretion" (Bradley et al. 1973).

"Larva: 16-22 mm (Meijerman and Ulenberg 2000), "Head shining black; prothoracic plate dark brown or black, edged with white anteriorly and sometimes posteriorly, divided by a narrow whitish line; abdomen whitish grey varying to dark bluish grey, paler or whitish laterally; pinacula light grey; setae whitish; anal plate black or blackish brown; anal comb present; thoracic legs black; prolegs green dotted with black" (Bradley et al. 1973).

Fig. 3. Larva of *Archips xylosteanus*
[Image UGA1297014, www.forestryimages.org]

Pupa: 11-12 mm (Beeke and De Jong 1991) and "dark brown or black" (Bradley et al. 1973).

Archips xylosteanus has a similar appearance to *Archips crataegana* (also not known to occur in the United States) but is generally smaller and more variegated (Bradley et al. 1973).

Fig. 4. Adult (left) and pupa (right) of *Archips xylosteanus*
[Image from Federal Research Station of Changins,
http://www.hortnet.co.nz/key/keys/bugkey2a/wings/dblwing/axylad1.htm]

A dichotomous key of common leafroller pests (Tortricidae) in larval and pupal stages is provided by Beeke and De Jong (1991).

Biology and Ecology

Archips xylosteanus has one generation per year (Dickler 1991). The insect overwinters in the egg stage (Razowski 1977, Benz 1991), and hatch begins in early spring (i.e., March) when host plants produce new growth on branches (Razowski 1977, Dickler 1991). Newly hatched larvae crawl to green branch tips and begin feeding. Later instars roll leaves diagonally and feed in seclusion (Razowski 1977). Collectively, larval development requires 30-40 days, but the pupal stage only lasts 9-12 days (Razowski 1977). Moths are active from late June or early July to mid August in Europe and Japan (Razowski 1977). Moths rest in foliage during the daytime and fly at night or when disturbed (Bradley et al. 1973). One female may deposit 200-3000 eggs in 5-8 egg masses (Razowski 1977).

Temperature thresholds for development or flight have not been reported in current, available literature.

See ‘Known Hosts’ for a listing of plants that can be attacked by *A. xylosteanus* and ‘Pest Importance’ for a discussion of its potential economic impact.

Pest Importance

Archips xylosteanus is not known to occur in the US and is a quarantine pest of concern. This insect was recently detected in Newfoundland, Canada, a first report for North America (Spears 2006).

Larvae are polyphagous feeders of ornamental trees and shrubs, particularly new foliage and buds (see ‘Known Hosts’) (Dickler 1991). Population size can vary from year to year, however damage is usually not severe or economically important (Razowski 1977, Dickler 1991, Özbek and Calmasur 2005). In its native range, the insect is attacked by several natural enemies which may hold populations in check (Miczulski and Koślińska 1976). Thus, the potential economic impact in the US in the absence of natural enemies is uncertain.

Risks associated with *A. xylosteanus* have not been evaluated formally in the past. Because the insect feeds on foliage, it is unlikely to be moved with fresh commodities for consumption (e.g., fresh fruit) (Spears 2006). The insect is more likely to be introduced in infested nursery stock (Dunkle 2006).

Symptoms

Archips xylosteanus larvae may cause significant defoliation by feeding on foliage and buds of deciduous trees and shrubs (Spears 2006). Developing larvae will roll leaves to create protected feeding sites.

Fig. 5. Oak leaf rolled by *Archips xylosteanus*
[Image from www.forestryimages.org]

Known Hosts

The larvae of *A. xylosteanus* feed on the foliage of numerous trees and woody plants:

Hosts	References
Abies sp. (fir)	(Bradley et al. 1973, Zhang 1994)
Acer sp. (maple)	(Bradley et al. 1973)
Citrus sp.	(van der Geest et al. 1991)
Corylus sp. (hazelnut)	(Bradley et al. 1973)
Fraxinus sp. (ash)	(Bradley et al. 1973)
Hypericum sp.	(Bradley et al. 1973)
Lonicera sp. (honeysuckle)	(Bradley et al. 1973)
Malus domestica (apple)	(Hwang 1974, Miczulski and Koślińska 1976, Zhang 1994)
Malus pumila (paradise apple)	(CAB 2006)
Prunus apetala (wild cherry)	(Konno 2005)
Prunus armeniaca (apricot)	(CAB 2006)
Prunus avium (sweet cherry)	(Safonkin 1998, CAB 2006)
Prunus grayana (wild cherry)	(Konno 2005)
Prunus persica (peach)	(Hrdý et al. 1979, CAB 2006)
Prunus verecunda (wild cherry)	(Konno 2005)

Hosts	**References**
Prunus sp. (cherry, plum)	(Zhang 1994)
Pyrus sp. (pear)	(Zhang 1994)
Quercus borealis (red oak)	(Tomić and Mihajlović 1979)
Quercus ilex (holly oak)	(Toimil 1987, CAB 2006)
Quercus pyrenaica (black oak)	(CAB 2006)
Quercus robur (common oak)	(CAB 2006)
Quercus sp. (oak)	(Bradley et al. 1973, Zhang 1994)
Rhododendron sp.	(Zhang 1994)
Rubus sp. (raspberry)	(Bradley et al. 1973, Zhang 1994)
Rosa canina (dog rose)	(CAB 2006)
Rosa sp.	(Özbek and Calmasur 2005)
Solanum sp. (nightshade)	(CAB 2006)
Tilia sp. (basswood)	(Bradley et al. 1973)
Ulmus sp. (elm)	(Bradley et al. 1973)
Ornamental trees and shrubs (nursery stock)	(Dunkle 2006)

Known Distribution

Archips xylosteanus has been reported from:

Location	**References**
Asia Minor	(Bradley et al. 1973)
Bulgaria	(Zhang 1994)
Canada (Newfoundland)	(Dunkle 2006)
China	(Bradley et al. 1973, Hwang 1974, Zhang 1994)
Europe	(Bradley et al. 1973)
France	(Zhang 1994)
Germany	(Zhang 1994)
Japan	(Bradley et al. 1973, Zhang 1994, Konno 2005)
Korea	(Razowski 1977)
Lithuania	(Zhang 1994)
Netherlands	(Beeke and De Jong 1991)
Poland	(Koślińska 1973, Miczulski and Koślińska 1976, Zhang 1994)
Romania	(Zhang 1994)
Russia	(Bradley et al. 1973, Zhang 1994, Safonkin 1998)
Serbia	(Tomić and Mihajlović 1979)
Slovakia	(Hrdý et al. 1979)

Location	References
Spain	(Toimil 1987, CAB 2006)
Sweden	(Zhang 1994)
Turkey	(Zhang 1994, Özbek and Calmasur 2005)
Ukraine	(Zhang 1994)
United Kingdom	(Bradley et al. 1973, Zhang 1994)

Potential Distribution within the US

Archips xylosteanus is a Palearctic species. Based on the list of countries In Europe and Asia from which the species has been reported, Schall (2006) predicts the species is likely to occur in regions (zonobiomes) with climates characterized as warm-temperate, typical-temperate, arid-temperate, and transitional to cold-temperate or boreal. Consequently, using this approach most of the contiguous US is predicted to be climatically suitable, with the exception of southern Florida, southern Texas, the desert southwest, and California's coast and Central Valley (Schall 2006).

Our own analysis of the reported geographic distribution of *A. xylosteanus* gives a similar, albeit slightly more restricted, prediction. Our analysis suggests that this insect is most likely to be associated with biomes defined by Olson et al. (2001) as temperate-broadleaf-and-mixed forest and boreal forest. Boreal forest does not occur in the US. Temperate-broadleaf-and-mixed forest is the most common biome east of the Mississippi River and accounts for 28% of the area within the contiguous US.

Survey

Male *A. xylosteanus* are attracted to blends of Z-11-tetradecenyl acetate and *E*-11- tetradecenyl acetate (El-Sayed 2006). Ando et al. (1978) were the first to demonstrate that male moths were attracted to a 4:1 mixture of *Z*-11-tetradecenyl acetate: *E*-11-tetradecenyl acetate, but captures with this blend were relatively low (only 13 moths over an unspecified length of time). This mixture also attracted the tortricids *Archippus piceanus similis* Butler and *Pandemis cinnamomeana* Treitschke (Ando et al. 1978). Frerot et al. (1979, 1983) found that the same two compounds in a 92:8 [*Z*:*E*] mixture captured substantially more male *A. xylosteanus* than any other ratio tested (approximately 150 males over an unspecified length of time). This ratio of these compounds may also be attractive to *Cacaecimorpha pronubana* Hübner and *Argyrotaenia pulchellana* Haw (Frerot et al. 1979). Conversely, *A. xylosteanus* may be attracted to pheromone lures for oriental fruit moth, *Grapholita molesta* (93:7 *Z*-8-dodecenyl acetate:E-8-dodecenyl acetate + docecanol), red-banded leafroller, *Argyrotaenia velutinana* (2:3 Z-11-tetradecenyl acetate:dodecyl acetate), and the oblique banded leafroller, *Choristoneura rosaceana* (Z-11-tetradecenyl acetate) (Hrdý et al. 1979).

Pheromones produced by *Archips rosana* may interfere with attractants for *A. xylosteanus* (Safonkin 1998).

Pheromone traps should be placed approximately 1.6 m [5 ft] above the ground and 50-100 m [150-300 ft] apart (Hrdý et al. 1979, Frerot et al. 1983). Pherocon 1C traps are more effective at capturing males than Stuttgart pot traps (Hrdý et al. 1979).

Individual species of leafrollers are difficult to detect with visual inspections of foliage. Leaf rolling is common among many tortricids, and *A. xylosteanus* may closely resemble related species. *Archips xylosteanus* is the type species for the Xylosteana group (Razowski 1997). Other introduced and native members of the Xylosteana group in North America include *A. argyrospila*, *A. cerasivorana*, *A. eleagnana*, *A. fervidana*, *A. fuscocupreana* (introduced), *A. georgiana*, *A. goyerana*, *A. grisea*, *A. infumatana*, *A. magnoliana*, *A. mortuana*, *A. myricana*, *A. negundana*, *A. nigriplagana*, *A. purpurana*, *A. rileyana*, *A. rosana* (introduced), and *A. semiferana* (Kruse and Sperling 2002). Like *A. xylosteanus*, *A. fervidana* and *A. semiferana* (Fig. 6) feed on oak. Both native species are common in the US.

Fig. 6. Adult *Archips fervidana* (upper) and *A. semiferana* (lower). Images not to scale.
[Images from T.M. Gilligan, Academy of Natural Sciences, Philadelphia, PA, www.tortricidae.com]

This insect can be difficult to identify, so any identification should be confirmed by an appropriately trained entomologist (Bradley et al. 1973, Beeke and De Jong 1991). Dichotomous keys for later instars and pupae of common leafrollers are provided by Beeke and De Jong (1991).

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Epiphyas postvittana[*]

Scientific Name

Epiphyas postvittana (Walker)

<u>Synonyms:</u>

At the generic level:

Epiphyas Turner 1927, Pap. Roy. Soc. Tasmania 1926: 125. Type species: *Epiphyas eucyrta* Turner, 1926.

- *Austrotortrix* Bradley, 1956, Bull. Entomol. Res. 47: 101. Type species: *Teras postvittana* Walker, 1863.
- *Austerotortrix* Razowski, 1977, [misspelling of *Austrotortrix.*]

At the species level:

postvittana Walker, 1863 (*Teras*), List Spec. Lepid. Ins. Colln. Brit. Mus. 28: 297. TL: Australia (Sydney). HT (♀): BMNH.

- *scitulana* Walker, 1863 (*Teras*), List. Spec. Lepid. Ins. Colln.. Brit. Mus. 28: 298. TL: Australia (Sydney). HT (♂): BMNH.
- *basialbana* Walker, 1863 (*Teras*), List Spec. Lepid. Ins. Colln. Brit. Mus. 28: 299. TL: Australia. HT (♂): BMNH.
- *secretana* Walker, 1863 (*Teras*), List Spec. Lepid. Ins. Colln. Brit. Mus. 28: 300. TL: Australia. HT (♀): BMNH.
- *consociana* Walker, 1863 (*Pandemis*), List Spec. Lepid. Ins. Colln. Brit. Mus. 28: 311. TL: Australia (Sydney). HT (♀): BMNH.
- *reversana* Walker, 1863 (*Dichelia*), List Spec. Lepid. Ins. Colln. Brit. Mus. 28: 321. TL: Australia (Sydney). HT (♂): BMNH.
- *foedana* Walker, 1863 (*Dichelia*), List Spec. Lepid. Ins. Colln. Brit. Mus. 28: 321. TL: Australia. HT (♀): BMNH.
- *retractana* Walker, 1863 (*Dichelia*), List Spec. Lepid. Ins. Colln. Brit. Mus. 28: 322. TL: Australia. HT (♀): BMNH.
- *vicariana* Walker, 1869 (*Dichelia*), Char. Undescr. Heter.: 82. TL: Australia. HT: NMVM.
- *stipularis* Meyrick, 1910 (*Tortrix*), Proc. Linnean Soc. N.S. Wales 35: 226. TL: Australia (Victoria, Murtoa). HT (♂): Lyell Collection.
- *pyrrhula* Meyrick, 1910 (*Tortrix*), Proc. Linnean Soc. N.S. Wales 35: 226. TL: Australia (South Australia, Port Lincoln). LT: BMNH.
- *oenopa* Meyrick, 1910 (*Tortrix*), Proc. Linnean Soc. N.S. Wales 35: 230. TL: Australia (Victoria). HT (♂): Lyell Collection.
- *dissipata* Meyrick, 1922 (*Tortrix*), Exotic Microlepid. 2: 496. TL: Australia (Yallingup). HT: BMNH.

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[*] This document is largely excerpted from the report: Venette, R.C., E.E. Davis, M. DaCosta, H. Heisler, and M. Larson. 2003. Mini-Risk Assessment: Light Brown Apple Moth, *Epiphyas postvittana* (Walker) [Lepidoptera: Tortricidae]. Available on-line at www.aphis.usda.gov/ppq/ep/pestdetection/pra/epostvittanapra.pdf

- *phaeosticha* Turner, 1939 (*Tortrix*), Pap. Proc.. Roy. Soc. Tasmania 1938: 76. TL: Tasmania. HT: Unknown.
- *vicaureana* Bradley, 1957 (*Dichelia*), Bull. Entomol. Res. 47: 103. [misspelling of *vicariana*].

(Taxonomic history provided by John Brown, National Museum of Natural History, pers. comm.)

Common Names

Light brown apple moth

Type of Pest

Moth, leafroller, defoliator

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for inclusion in manual

CAPS Priority Pest (FY 2007)

Pest Description

Fig. 1. Life stages of *Epiphyas postvittana*: (top left) eggs; (top right) larva; (bottom left) pupa, (bottom right) adult male [left] and female [right].
[Images from http://www.hortnet.co.nz/key/keys/info/lifecycl/lba-desc.htm]

“Male 16-21 mm, female 17-25 mm. Sexual dimorphism pronounced; male usually smaller, antenna weakly dentate-ciliate, ... basal half of forewing usually sharply demarcated, well-developed costal fold from base to about two-fifths; ... The male is usually distinguished by the abrupt division of the forewing medially into a pale basal area and darker apical area, and the female by its large size and relatively elongate forewing, often with greatly reduced markings” (Bradley 1973).

Fig. 2. Dorsal views of *Epiphyas postvittana* (Walker), A-male, B-female, C-male, D-female
[Reproduced from Bradley et al. (1979)]

The female abdomen is yellowish ash-colored (Hampson 1863).

Male wings: “Basal half of forewing light buff or pale yellow, contrasting strongly with the dark brown and rusty red-brown coloration of the distal half, the demarcation often emphasized by the deeper coloration of the oblique, narrow median fascia, the inner edge of which is sharply defined and usually straight, but sometimes is slightly wavy at the middle; pre-apical spot obscure, its inner margin usually defined by rusty red-brown ground coloration separating it from the median fascia. Hindwing gray ... *Epiphyas postvittana* (Walker) is extremely variable with numerous recurring forms. In strongly marked forms of the male the distal half of the forewing may vary from reddish brown to blackish, often with purplish mottling; the contrasting pale basal half may be sparsely speckled with black. Lightly marked forms resembling the female in appearance occur; an extreme form in which the usually dark outer half of the forewing is light and the pre-apical spot discernible is uncommon (Fig. 2 A and C) “(Bradley 1973).

Female wings: “General coloration of the forewing more uniform, with less contrast between the basal and distal halves; median fascia usually reduced. ... Only minor variation is found in the female; often the forewing is irrorate with black in both the basal and distal halves of the wing” (Fig. 2 B and D) (Bradley 1973).

More detailed, technical descriptions of the morphology of *E. postvittana* are provided by Zimmerman (1978), Hampson (1863), Bradley et al (1973) and Scott (1984).

Biology and Ecology

In much of Australia, *E. postvittana* completes three generations annually (Danthanarayana 1975, Geier and Briese 1980, Thomas 1989). Cooler temperatures lead to longer development times for all stages of growth (Magarey et al. 1994). In summer the life cycle takes 4-6 weeks to complete (Nuttal 1983), but more than three generations can be completed if temperatures and host plants are favorable (MacLellan 1973, Thomas 1989, Madge and Stirrat 1991, Bailey 1997). For example, four generations can be completed in southeastern Australia where it is warmer (Buchanan et al. 1991, Magarey et al. 1994). In contrast, two generations occur in Tasmania (Evans 1937), New Zealand (McLaren and Fraser 1992), and the UK (Bradley 1973). In Australia, generations do not overlap, but they do in the UK (Bradley 1973). Within a generation several life stages of the insect (e.g., eggs and larvae; Fig. 1) may co-occur (Danthanarayana 1975).

Adult moths emerge after one to several weeks of pupation (Magarey et al. 1994). Female moths emerge from protective pupal nests (see below) and mate soon after emergence (Geier and Briese 1981). Danthanarayana (1975) suggests the preoviposition period is 2-7 days. Females copulate for slightly less than 1 hr (Foster et al. 1995). Oviposition does not begin until females are 2- to 3-days old (Geier and Briese 1981). The oviposition period lasts 1-21 days (Danthanarayana 1975). Adult longevity is influenced by host plant and temperature. In the laboratory, female longevity can vary between 10 days (Geier and Briese 1981) and 32.7 days (Danthanarayana 1975); males can live up to approximately 33 days (Danthanarayana 1975). In the field in Australia, the life span of adult *E. postvittana* is 2-3 weeks (Magarey et al. 1994). Heavier females live longer and lay more eggs than lighter females (Danthanarayana 1975). Female moths are typically larger than males (Danthanarayana 1975, Geier and Briese 1981).

Moths are quiescent during the day and may be found on foliage of hosts (Geier and Briese 1981). Flight occurs at dusk in calm conditions (Geier and Briese 1981, USDA 1984, Magarey et al. 1994). Females deposit eggs at night (USDA 1984). Adults are unlikely to disperse from areas with abundant, high-quality hosts (Geier and Briese 1981). Males will disperse farther than females. In a mark-release-recapture study, 80% of recaptured males and 99% of recaptured females occurred within 100 m of the release point (Suckling et al. 1994). Females do not appear to rely on plant volatiles to locate a host, but tactile cues are important (Foster and Howard 1998). Humidity influences the dispersal ability of the pest (Danthanarayana et al. 1995).

Females deposit eggs in egg masses. Within a mass, eggs are “stuck together like roof tiles” (Fig. 1) (Geier and Briese 1981) and are covered in a greenish “waxy secretion” (Evans 1937, Nuttal 1983). The number of eggs deposited in a mass is variable. Typically, females deposit 20 to 50 eggs per mass (Danthanarayana 1975, Geier and Briese 1981, Nuttal 1983, USDA 1984, Magarey et al. 1994). A female moth may produce up to 1,492 eggs (Danthanarayana 1975, 1983), but the average number of eggs produced per female typically varies between 118-462 (MacLellan 1973, Danthanarayana 1975, Geier and Briese 1981, USDA 1984, Danthanarayana et al. 1995). Fecundity is greatest at temperatures between 20 and 25°C [68-77°F], inclusive (Danthanarayana et al. 1995). Females prefer smooth leaf surfaces on which to deposit their eggs (Danthanarayana 1975, Geier and Briese 1981, Foster and Howard 1998). The egg stage lasts an average of 5-7 days at a temperature of 28°C [82°F] (Danthanarayana 1975). Egg-hatching ceases at temperatures greater than 31.3°C [88°F] (Danthanarayana 1975).

Epiphyas postvittana typically completes five to seven instars (Danthanarayana 1975, Geier and Briese 1981, Magarey et al. 1994). Larvae emerge from eggs after 1-2 weeks and disperse, usually to the underside of the leaf, where they spin a “silken shelter” (i.e., a silken tunnel) and commence feeding (Danthanarayana 1975, Geier and Briese 1981, Nuttal 1983, USDA 1984, Thomas 1989). Although they are sheltered in silk, first instar larvae are more exposed to weather and insecticide treatments than are second and third instar larvae (Madge and Stirrat 1991, Lo et al. 2000). After approximately 3 weeks, larvae leave the silken tunnels for a new leaf (USDA 1984). Second and later instars have the ability to create their own protective feeding shelter by rolling a leaf or webbing multiple leaves together (Danthanarayana 1975, Lo et al. 2000), behaviors characteristic of the Tortricidae.

Larvae move vigorously when disturbed but are always connected to the leaf by a silken thread to avoid being removed from a leaf (Nuttal 1983, USDA 1984). When larvae happen to fall to the ground, they feed on ground-cover hosts or can survive without feeding for several months (Evans 1937, Thomas 1975, USDA 1984).

In cold climates, larvae are the overwintering stage (Nuttal 1983). Larvae prepare to overwinter by locating “sheltering niches,” which may be mummified fruit or ground vegetation (Thomas 1975). Overwintering larvae can utilize alternate hosts, including several weed species, for food and to form shelters (Buchanan et al. 1991). Larvae may also survive winters without feeding for up to 2 months (USDA 1984). *Epiphyas postvittana* does not diapause (Geier and Briese 1981), rather, development is slowed under cold winter temperatures (MacLellan 1973, Geier and Briese 1981, Danthanarayana 1983, USDA 1984). Development is only likely to occur at temperatures between 7.1° and 30.7°C [45-87°F] (Danthanarayana et al. 1995).

Pupation is completed within the "nests" made from rolled-up leaves (Danthanarayana 1975, Geier and Briese 1981, Nuttal 1983, Magarey et al. 1994). The pupal stage lasts 2-3 weeks (Evans 1937).

Epiphyas postvittana is more abundant during the second generation than during other generations (MacLellan 1973, Madge and Stirrat 1991). Thus, the second generation causes the most economic damage (Evans 1937, Thomas 1975, Madge and Stirrat 1991, Lo et al. 2000) as larvae move from foliage to fruit (MacLellan 1973, Magarey et al. 1994). The size of the third generation is typically smaller than the previous two due to leaf fall (including attached larvae) as temperatures decline in autumn (Thomas 1975). The level of damage caused by *E. postvittana* is not related to the potential number of generations that the pest may complete (Geier and Briese 1981).

Several studies describe the developmental thresholds and accumulated degree days necessary for the completion of each life stage (Table D1). A phenological model developed with parameters from Danthanarayana (1975) and Geier and Springett (1976) performed better when the accumulation of degree-days [°C] began at "budburst" rather than at a start date of July 1 (Madge and Stirrat 1991). Although important discrepancies between the predicted and observed population dynamics were noted, the performance of the model was considered acceptable (Madge and Stirrat 1991).

Table 1. Developmental threshold and degree day requirements for *E. postvittana.*

Stage	Developmental threshold (˚C)	Degree Days ± SE	Notes	Reference
Egg	7.0	131 ± 1	Lab study	(Geier and Briese 1981)
	7.5	133.7	Lab study	(Danthanarayana 1975)
Larva	6.9	380.8 ± 13.2	Average over several host plants; from authors' Table 2	(Danthanarayana et al. 1995)
	7.5 lower, 31-32 upper	345.9	Lab study	(Danthanarayana 1975)
Pupa	3.8	175.0 ± 11.1	Average over several host plants; from authors' Table 2	(Danthanarayana et al. 1995)
	7	132 ± 2	Lab study	(Geier and Briese 1981)
	7.5 lower, 31-32 upper	129.1	Lab study	(Danthanarayana 1975)
Adult	-3.2	393.1 ± 9.4	Adult longevity; from authors' Table 3	(Danthanarayana et al. 1995)

Stage	Developmental threshold (˚C)	Degree Days ± SE	Notes	Reference
	6.9	NA	Female; lab study	(Geier and Briese 1981)
	7.1	NA	Male; lab study	(Geier and Briese 1981)
	7.5	29.9	Preoviposition period	(Danthanarayana 1975)
	7.5	83	Eclosion to 50% oviposition	(Danthanarayana 1975)
Neonate to pupa	7	265-551	Range influenced by host quality	(Geier and Briese 1981)
Complete life cycle	7.5	620.5	Egg to first egg	(Danthanarayana 1975)
	7.5	673.6	Egg to 50% oviposition	(Danthanarayana 1975)

Pest Importance

Epiphyas postvittana is a highly polyphagous pest that attacks a wide number of fruits and other plants. This species has a relatively restricted geographic distribution, being found only in portions of Europe and Oceania (van Den Broek 1975, Terauds 1977, IIE 1991, Danthanarayana et al. 1995, Suckling et al. 1998). *Epiphyas postvittana* is native to Australia but has successfully invaded other countries (Danthanarayana 1975).

The potential impact of this species on oak has not been well studied. Other forest/plantation species have received more attention. Conifers are damaged by needle-tying and chewing (Nuttal 1983). Larvae have been found feeding near apices of Bishop pine (*Pinus muricata*) seedlings where they spin needles down against the stem and bore into the main stem from the terminal bud (Winter 1985).

Epiphyas postvittana is reported as a pest of economic importance to many ornamental and fruit crops throughout its range (Zhang 1994). According to Geier (1981) "Economic damage results from feeding by caterpillars, which may:

- destroy, stunt or deform young seedlings…
- spoil the appearance of ornamental plants
- injure deciduous fruit-tree crops, citrus, and grapes".

Some varieties of apples such as 'Sturmer Pippin' (an early variety), 'Granny Smith' and 'Fuji' (late varieties) can experience up to 20% damage (Suckling and Ioriatti 1996), while severe attacks can damage 75%-85% of a crop (Danthanarayana 1975, USDA 1984). In 1992, a severe outbreak (70,000 larvae/ha) caused a loss of 4.7t of chardonnay fruit (Bailey et al. 1995). Damage in the 1992-93 Chardonnay season at Coonawarra (southern Australia) cost $2,000/ha (Bailey et al. 1996). Mature larvae are the most difficult stage to control (Lay-Yee et al. 1997). A single larva can destroy about 30 g [1 oz] of mature grapes (Bailey 1997).

Epiphyas postvittana is a difficult to control with sprays because of its leaf-rolling ability, and because there is evidence of resistance due to overuse of sprays (Geier and Briese 1981).

The likelihood and consequences of establishment by *E. postvittana* have been evaluated in pathway-initiated risk assessments. *Epiphyas postvittana* was considered highly likely of becoming established in the US; the consequences of its establishment for US agricultural and natural ecosystems were judged to be high (i.e., severe) (Lightfield 1995). Canada has listed *E. postvittana* as a noxious pest, and the presence of the pest would prevent export of any infested commodity (Danthanarayana et al. 1995). In New Zealand, the recommended economic threshold is six or more larvae per 30 m [98 ft] row of fruit crops, however if the crop is intended for export, control is recommended if only one larva is found (Charles et al. 1987).

Symptoms

The insect will feed on foliage, flowers, and fruit. In spring, the pest feeds on new buds while later generations feed on ripened fruits (Buchanan et al. 1991). “After the first moult they construct typical leaf rolls (nests) by webbing together leaves, a bud and one or more leaves, leaves to a fruit, or by folding and webbing individual mature leaves. During the fruiting season they also make nests among clusters of fruits, damaging the surface and sometimes tunneling into the fruits” (Danthanarayana 1975).

Feeding damage on oaks is not well described, but leaf rolling by later instar larvae provides an indication of the potential presence of the species. Other tortricids feed on oak and may roll leaves.

Feeding injury to fruit crops is typically caused by later instars (Lo et al. 2000). Fruits are not preferred feeding sites, so feeding there is thought to happen by chance (Geier and Briese 1980, Lo et al. 2000). However, volatiles emitted by ripening fruit may be attractive to larvae (Suckling and Ioriatti 1996). On a fruit, the calyx offers protection from parasitoids and is probably the best feeding location for young larvae (Lo et al. 2000). Damage to the host plant is compounded by the pest, as it acts as a “vector” to spread fungal disease; feeding injury also predisposes the host to fungal infection (Buchanan et al. 1991, Bailey et al. 1995, Bailey 1997, Lo et al. 2000).

Damage to apples is in the form of either pinpricks, which are flask-shaped holes about 3 mm deep into the fruit, or entries, which are holes extending deeper than 3 mm into the fruit that leaves some frass and webbing at the surface (van Den Broek 1975). The first generation (in spring) causes the most damage to apples while the second generation damages fruit harvested later in the season

(Terauds 1977). Peaches are damaged by feeding that occurs on the shoots and fruit (Lo et al. 1995).

Known Hosts

Epiphyas postvittana has a host range in excess of 120 plant genera in over 50 families (Geier and Briese 1981) with preferences for hosts in the families Compositae, Leguminosae, Polygonaceae, and Rosaceae (Danthanarayana 1975).

Hosts	**References**
Acacia spp. (wattle)	(Danthanarayana 1975, Geier and Briese 1981)
Actinidia chinensis (Chinese gooseberry)	(CAB 2003)
Actinidia deliciosa (kiwifruit)	(Stevens 1995)
Adiantum sp. (maidenhair fern)	(Geier and Briese 1981)
Alnus glutinosa (black alder/European alder)	(Suckling et al. 1998)
Amaranthus sp. (amaranth)	(Danthanarayana 1975, Geier and Briese 1981)
Aquilegia sp. (columbine)	(Geier and Briese 1981)
Arbutus sp. (madrone)	(Geier and Briese 1981)
Arctotheca calendula (capeweed)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Artemisia sp. (sagebrush)	(Geier and Briese 1981)
Astartea sp. (astartea)	(Geier and Briese 1981)
Aster sp. (aster)	(Danthanarayana 1975, Geier and Briese 1981)
Baccharis sp. (baccharis)	(Danthanarayana 1975, Geier and Briese 1981)
Boronia sp. (boronia)	(Danthanarayana 1975, Geier and Briese 1981)
Brassica sp. (mustard)	(Danthanarayana 1975, Geier and Briese 1981)
Breynia sp. (breynia)	(Geier and Briese 1981)
Buddleia spp. (butterfly bush)	(Danthanarayana 1975, Geier and Briese 1981, Zhang 1994)
Bursaria sp. (bursaria)	(Geier and Briese 1981)
Calendula sp. (marigold)	(Danthanarayana 1975, Geier and Briese 1981)
Callistemon sp. (bottlebrush)	(Geier and Briese 1981)
Camellia japonica (camellia), *Camellia* sp.	(Geier and Briese 1981)
Campsis sp. (campsis)	(Geier and Briese 1981)
Cassia sp. (cassia)	(Geier and Briese 1981)

Hosts	**References**
Ceanothus sp. (ceanothus)	(Geier and Briese 1981)
Centranthus spp. (fox's brush/heliotrope/valerian)	(Danthanarayana 1975, Geier and Briese 1981, Zhang 1994)
Chenopodium album (fat-hen)	(Danthanarayana 1975, Geier and Briese 1981)
Choisya sp. (Mexican orange)	(Geier and Briese 1981)
Chrysanthemum spp., *Chrysanthemum* x *morifolium* (chrysanthemum)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Citrus spp. (citrus)	(Danthanarayana 1975, Geier and Briese 1981, Nuttal 1983, IIE 1991, CAB 2003)
Clematis sp. (leather flower)	(Geier and Briese 1981)
Clerodendron sp. (clerodendron)	(Geier and Briese 1981)
Correa sp. (Australian fuschia)	(Danthanarayana 1975, Geier and Briese 1981)
Cotoneaster spp. (cotoneaster)	(Geier and Briese 1981, CAB 2003)
Crataegus spp. (hawthorn)	(Geier and Briese 1981, CAB 2003)
Crocosmia spp. (montbretia)	(Danthanarayana 1975)
Cupressus sp. (cyprus)	(Danthanarayana 1975, Geier and Briese 1981, Winter 1985)
Cydonia sp. (cydonia)	(Geier and Briese 1981)
Cytisus scoparius (Scotch broom)	(Tomkins et al. 1989, Zhang 1994, Suckling et al. 2001, CAB 2003)
Dahlia sp. (dahlia)	(Geier and Briese 1981)
Datura sp. (datura)	(Geier and Briese 1981)
Daucus sp. (wild carrot)	(Geier and Briese 1981)
Diospyros kaki (persimmon)	(IIE 1991, Dentener et al. 1996)
Diospyros spp. (malabar ebony)	(CAB 2003)
Dodonaea sp. (dodonaea)	(Geier and Briese 1981)
Eriobotrya sp. (loquat)	(Geier and Briese 1981)
Eriostemon sp. (waxflower)	(Geier and Briese 1981)
Escallonia sp. (redclaws)	(Danthanarayana 1975, Geier and Briese 1981, Zhang 1994)
Eucalyptus spp. (eucalyptus)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Euonymus spp. (euonymus)	(Danthanarayana 1975, Geier and Briese 1981, Zhang 1994)
Feijoa sellowiana (horn of plenty)	(Geier and Briese 1981, CAB 2003)
Forsythia sp. (forsythia)	(Geier and Briese 1981)
Fortunella sp. (kumquat)	(Geier and Briese 1981)

Hosts	References
Fragaria sp. (strawberry)	(Danthanarayana 1975, Geier and Briese 1981, IIE 1991)
Gelsemium sp. (trumpetflower)	(Geier and Briese 1981)
Genista sp. (broom)	(Geier and Briese 1981)
Gerbera sp. (Transvaal daisy)	(Geier and Briese 1981)
Grevillea sp. (grevillea)	(Danthanarayana 1975, Geier and Briese 1981)
Hardenbergia sp.	(Geier and Briese 1981)
Hebe spp. (hebe)	(Danthanarayana 1975, Geier and Briese 1981, Zhang 1994)
Hedera helix, *Hedera* spp. (ivy)	(Danthanarayana 1975, Geier and Briese 1981, Zhang 1994)
Helichrysum sp. (strawflower)	(Geier and Briese 1981)
Humulus lupulus (hop)	(CAB 2003)
Hypericum perforatum (St. John's wort)	(Danthanarayana 1975, Geier and Briese 1981, Zhang 1994)
Jasminum spp. (jasmine)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Juglans sp. (walnut)	(Danthanarayana 1975, Geier and Briese 1981, Suckling et al. 1998)
Lathyrus sp. (pea)	(Geier and Briese 1981)
Lavendula sp. (lavender)	(Danthanarayana 1975, Geier and Briese 1981)
Leptospermum sp. (teatree)	(Geier and Briese 1981)
Leucadendron sp.	(Danthanarayana 1975, Geier and Briese 1981)
Ligustrum vulgare, *Ligustrum* spp. (privet)	(Danthanarayana 1975, Geier and Briese 1981, Zhang 1994, CAB 2003)
Linus sp.	(Geier and Briese 1981)
Litchi chinensis (litchi)	(Geier and Briese 1981, CAB 2003)
Lonicera sp. (honeysuckle)	(Geier and Briese 1981)
Lupinus sp. (lupine)	(Danthanarayana 1975, Geier and Briese 1981)
Lycopersicum sp. (tomato)	(Danthanarayana 1975, Geier and Briese 1981)
Macadamia sp. (macadamia)	(Geier and Briese 1981)
Malus domestica, *Malus* spp. (apple)	(Danthanarayana 1975, Terauds 1977, Tomkins et al. 1989, Zhang 1994, Danthanarayana et al. 1995, Suckling et al. 2001, CAB 2003)
Mangifera sp. (mango)	(Geier and Briese 1981)

Hosts	**References**
Medicago sativa (lucerne/alfalfa)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Melaleuca sp. (melaleuca)	(Geier and Briese 1981)
Mentha sp. (mint)	(Danthanarayana 1975, Geier and Briese 1981)
Mesembryanthemum sp. (iceplant)	(Danthanarayana 1975, Geier and Briese 1981)
Michelia sp. (michelia)	(Geier and Briese 1981)
Monotoca sp.	(Geier and Briese 1981)
Myoporum sp. (myoporum)	(Geier and Briese 1981)
Oxalis sp. (woodsorrel)	(Geier and Briese 1981)
Parthenocissus sp. (creeper)	(Geier and Briese 1981)
Pelargonium sp. (geranium)	(Geier and Briese 1981)
Persea americana (avocado)	(CAB 2003)
Persoonia sp. (geebungs/snottygobbles)	(Danthanarayana 1975, Geier and Briese 1981)
Petroselinum sp. (parsley)	(Geier and Briese 1981)
Philadelphus sp. (mock orange)	(Geier and Briese 1981)
Photinia sp. (chokeberry)	(Geier and Briese 1981)
Pinus muricata, *P. radiata*, *Pinus* spp. (pine)	(Danthanarayana 1975, Geier and Briese 1981, Winter 1985, Brockerhoff et al. 2002, CAB 2003)
Pittosporum sp. (cheesewood)	(Geier and Briese 1981)
Plantago lanceolata (plantain/ribwort)	(Danthanarayana 1975, Geier and Briese 1981, Tomkins et al. 1989, Zhang 1994, Danthanarayana et al. 1995, Suckling et al. 1998, CAB 2003)
Platysace sp.	(Geier and Briese 1981)
Polygala sp. (polygala)	(Geier and Briese 1981)
Polygonum sp. (knotweed)	(Danthanarayana 1975, Geier and Briese 1981)
Populus nigra (black poplar), *Populus* spp.	(Tomkins et al. 1989, Zhang 1994, Suckling et al. 1998, Suckling et al. 2001, CAB 2003)
Prunus armeniaca (apricot)	(Danthanarayana 1975, Whiting and Hoy 1997, CAB 2003)
Prunus persica (peach)	(Danthanarayana 1975, Zhang 1994, Lo et al. 1995, Brown and Il'ichev 2000, CAB 2003)
Pteris sp. (brake fern)	(Geier and Briese 1981)
Pulcaria sp.	(Danthanarayana 1975, Geier and Briese 1981)

Hosts	**References**
Pyracantha sp. (firethorn)	(Danthanarayana 1975, Geier and Briese 1981)
Pyrus spp. (pear)	(Danthanarayana 1975, Geier and Briese 1981, Nuttal 1983, CAB 2003)
Quercus spp. (oak)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Ranunculus sp. (buttercup)	(Geier and Briese 1981)
Raphanus sp. (radish)	(Danthanarayana 1975, Geier and Briese 1981)
Reseda sp. (mignonette)	(Danthanarayana 1975, Geier and Briese 1981)
Ribes spp. (currant)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Rosa spp. (rose)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Rubus idaeus (raspberry), *Rubus* spp. (blackberry/boysenberry/olallieberry)	(Danthanarayana 1975, Geier and Briese 1981, Charles et al. 1987, Tomkins et al. 1989, IIE 1991, Zhang 1994, Charles et al. 1996, CAB 2003)
Rumex crispus (curled dock)	(Danthanarayana 1975, Danthanarayana et al. 1995, CAB 2003)
Rumex obtusifolius (broadleaf dock)	(Tomkins et al. 1989, Zhang 1994, Suckling et al. 1998, CAB 2003)
Salix spp. (willow)	(Suckling et al. 1998, CAB 2003)
Salvia sp. (sage)	(Geier and Briese 1981)
Senecio sp. (ragwort)	(Geier and Briese 1981)
Sida sp. (fanpetals)	(Geier and Briese 1981)
Sisymbrium sp. (hedgemustard)	(Danthanarayana 1975, Geier and Briese 1981)
Smilax sp. (greenbrier)	(Geier and Briese 1981)
Solanum tuberosum (potato)	(Danthanarayana 1975, CAB 2003)
Sollya sp. (sollya)	(Geier and Briese 1981)
Tithonia sp. (tithonia)	(Danthanarayana 1975, Geier and Briese 1981)
Trema sp. (trema)	(Geier and Briese 1981)
Trifolium repens, *Trifolium* spp. (clover)	(Danthanarayana 1975, Geier and Briese 1981, Burnip and Suckling 1997) (Danthanarayana et al. 1995, Suckling et al. 1998, Suckling et al. 2001, CAB 2003)
Triglochin sp. (arrowgrass)	(Geier and Briese 1981)

Hosts	References
Ulex europaeus (gorse)	(Danthanarayana 1975, Geier and Briese 1981, Tomkins et al. 1989, CAB 2003)
Urtica sp. (nettle)	(Danthanarayana 1975, Geier and Briese 1981)
Vaccinium sp. (blueberry)	(Geier and Briese 1981, IIE 1991, CAB 2003)
Viburnum sp. (viburnum)	(Geier and Briese 1981)
Vicia faba (broad bean)	(Danthanarayana 1975, Geier and Briese 1981, CAB 2003)
Vinca sp. (periwinkle)	(Geier and Briese 1981)
Vitis vinifera, *Vitis* spp. (grape)	(Danthanarayana 1975, Geier and Briese 1981, IIE 1991, Glenn and Hoffmann 1997, CAB 2003)

Known Distribution

Epiphyas postvittana is found in northern Europe, southern Australia, New Zealand, and Hawaii (IIE 1991).

Location	References
Australia	(IIE 1991)
England	(IIE 1991, CAB 2003)
Hawaii	(IIE 1991)
New Caledonia	(IIE 1991)
New Zealand	(IIE 1991)

Potential Distribution within the US

Epiphyas postvittana performs best under cool conditions (mean annual temperature of ~13.5°C [56°F]) with moderate rainfall (~750 mm [29½ in] annually) and moderate-high relative humidity (~70%) (Danthanarayana et al. 1995). Hot, dry conditions may nearly eliminate a population (Danthanarayana 1983). The climate within its range can be generally characterized as temperate, tropical, or dry (CAB 2003). The currently reported global distribution of *E. postvittana* suggests that the pest may be most closely associated with deserts and xeric shrubland (likely where irrigated); temperate broadleaf and mixed forests; temperate grasslands, savannahs, and shrublands; and tropical and subtropical moist tropical broadleaf forests. Consequently, we estimate that approximately 80% of the contiguous US may be climatically suitable for *E. postvittana.*

Survey

Visual inspections have been used to monitor population dynamics of *E. postvittana* eggs and larvae. In grape, 40 vines were inspected per sampling date (Buchanan et al. 1991). In apple and other tree fruits, 200 shoots and 200 fruit clusters (10 of each on 20 different trees) are often inspected (Bradley et al.

1998, Lo et al. 2000). Egg masses are most likely to be found on leaves (USDA 1984). Larvae are most likely to be found near the calyx or in the endocarp; larvae may also create “irregular brown areas, rounds pits, or scars” on the surface of a fruit (USDA 1984). Larvae may also be found inside furled leaves, and adults may occasionally be found on the lower leaf surface (USDA 1984).

A sex pheromone has been identified from *E. postvittana* and used to monitor male flight periods. Two key components of the pheromone are (*E*)-11-tetradecenyl acetate and (*E*,*E*)-(9,11)-tetradecadienyl acetate (Bellas et al. 1983). These compounds in a ratio of 20:1 are highly attractive to males (Bellas et al. 1983). To monitor male flight activity in stands of Monterey pine (*Pinus radiata*) in New Zealand, 100 µg of a 95:5 ratio of (*E*)-11-tetradecenyl acetate: (*E*,*E*)-(9,11)- tetradecadien-1-yl acetate was placed on a rubber septum and used in delta traps with a 20 cm x 20 cm sticky base (Brockerhoff et al. 2002). Traps were placed 2 m [ca. 6.5 ft] above ground level without any understory vegetation (Brockerhoff et al. 2002). A similar procedure has been used in apples (Thomas and Shaw 1982, Suckling et al. 1990, Suckling and Shaw 1992, Bradley et al. 1998) and caneberries (e.g., raspberries and blackberries, Charles et al. 1996). Delta traps were placed 1.5 m [5 ft] above the ground, and lures were changed every 6 weeks (Thomas and Shaw 1982, Suckling et al. 1990, Suckling and Shaw 1992).

For a regional survey of tortricids, delta traps (20x20 cm sticky, flat base) were placed in each of 12 apple orchards (Cross 1996). Delta traps have also been used with pheromone lures to monitor male flights of *E. postvittana* in stone fruits (Brown and Il'ichev 2000). Frequently, traps are placed in the center of an orchard at densities in the range of 1 trap per 0.37-5 acres [=0.14-2 ha] (Bradley et al. 1998). In vineyards, pheromone traps also have been placed at a density of approximately 1 trap per 5 acres (2 ha) (Glenn and Hoffmann 1997).

Foster and Muggleston (1993) provide a detailed analysis of different designs of delta traps. In general, they found that traps with a greater length (i.e., the distance between the two openings of the trap) capture significantly more *E. postvittana* than shorter traps. This effect is not related to saturation of smaller sticky surfaces with insects or other debris. The addition of barriers to slow the exit of an insect from a trap also improves catch. In a separate analysis, Foster et al. (1991) found that placing the pheromone lure on the side of the trap helped to improve trap efficiency. The orientation of the trap relative to wind direction did not affect the number of *E. postvittana* that were attracted to the pheromone or were subsequently caught by the trap (Foster et al. 1991).

Adults are also attracted to fruit fermentation products as a 10% wine solution has been used as an attractant and killing agent for adults (Buchanan 1977, Glenn and Hoffmann 1997). The dilute wine (670 ml [~23 oz]) in 1 liter jars was hung from grapevines on the edge of a block of grapes (Buchanan 1977).

Blacklight traps have been used to monitor adults of *E. postvittana* (Thwaite 1976).

Epiphyas postvittana may be confused with *E. pulla* [not known in US] and *E. liadelpha* [not known in US], and larvae of several leafrollers within its range (CAB 2003). Identity of the species must often be confirmed by examination of adult genitalia. Molecular diagnostics based on PCR amplification of ribosomal DNA have been developed and are especially useful for the identification of immature specimens (Armstrong et al. 1997). A morphological key to the larvae and pupae of *Epiphyas postvittana* (Walker) and *Amorbia emigratella* Busck, another tortricid that looks like *E. postvittana*, is provided in Zimmerman (1978).

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Erannis defoliaria

Scientific Name

Erannis defoliaria (Clerck)

Two subspecies are recognized (Scoble 1999):
Erannis defoliaria defoliaria Clerck
Erannis defoliaria teriolensis Dannehl

Synonyms:
Biston defoliaria Clerck
Hibernia defoliaria Clerck
Hybernia defoliaria Clerck
Phalaena defoliaria Clerck
Phalaena pulveraria Linnaeus
Phalaena geometra
Erannis defoliaria defoliaria Clerck
Erannis bistrigaria Giorna
Erannis defoliaria suffusa Cockerell
Erannis discolor Ström
Erannis pulveraria Hufnagel

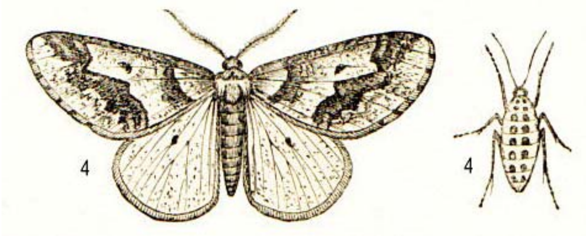

Fig. 1. Line drawing of male (left) and female (right) *Erannis defoliaria.*
[Image from L. Watson and M.J. Dallwitz, http://delta-intkey.com/britin/images/apter01.jpg]

Common Name

Mottled umber moth

Type of Pest

Moth, defoliator

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera,
Family: Geometridae, **Subfamily:** Ennominae

Reason for inclusion in manual

Exotic Forest Pest Information System – classified as a very high risk pest with the potential to attack oaks

Pest Description

Formally, the members of the genus *Erannis* are distinguishable by the following characters:

"Adults. Crepuscular to nocturnal; with fully developed wings, or flightless. The flightless females apterous (the body light grey with contrast black markings). Antennae of males shortly bipectinate (the short pectinations fasciculate-ciliate); apically simple; fasciculate-ciliate. Face rough (haired)" (Watson and Dallwitz 2003).

"Wingspan 40–45 mm [1.5-2 in]. The outer margin of the forewing convexly curved to more or less straight. Forewings whitish-ochreous, strigulated dark fuscous, with reddish-ochreous, ferrugineous, darkly fuscous or purplish-fuscous basal and post-median bands; or dark ochreous to ferrugineous, darkly speckled and/or strigulated; with a clear discal mark (usually, this blackish), or without a clear discal mark. Forewings of the male without a fovea. Hindwings somewhat conspicuously patterned to plain; whitish-ochreous, sprinkled grey; with a clear discal mark (this dark); transversely lined to without transverse lines" (Watson and Dallwitz 2003).

"Hindwings lacking a tubular vein 5. Vein 8 of the hindwings approximated to or anastomosed with the upper margin of the cell to the middle or beyond. Hindwing veins 6 and 7 separate" (Watson and Dallwitz 2003).

"Thorax crested. Posterior tibiae of males 4-spurred.

The abdomen conspicuously patterned (grey with black spots, in the female), or plain (in the male)" (Watson and Dallwitz 2003).

Larvae of *E. defoliaria* are "28-32 mm [1.1-1.25 in long]. Body greyish, heavily marked with reddish-brown, with a wavy black subdorsal line that has pale yellow blotches below. Head reddish-brown and the white spiracles ringed with black" (Porter 1997).

Fig. 2. Larva of *Erannis defoliaria* on an oak leaf.
[Image from Milan Zubrick, www.forestryimages.org,
image UGA1370060]

Coloration of adult *E. defoliaria* (particularly the ground color of wings) varies considerably from dark grey to buff (Skinner 1984). In general, male moths have "pale-brown or brownish-yellow forewings, banded and mottled with dark brown. The female is wingless, the yellow brown body having two more spots on every segment" (Maff 1978). Adults look much like *E. tiliaria*, present in North America (Rindge 1975); see 'Survey'. A formal description of *E. defoliaria* was not readily available, but adult male *E. tiliaria* have "head with vertex yellowish brown to pale brown; front brown to grayish brown; palpi grayish brown to dark brown. Thorax above yellowish brown to pale brown, with grayish white scaling on patagia and posterior tuft; below grayish brown anteriorly, pale gray posteriorly; legs with mixed gray, brown, and blackish brown scales, tarsi with ends of segments narrowly grayish white. Abdomen above yellowish brown with scattered dark brown scales; below pale grayish brown with scattered dark brown scales" (Rindge 1975).

"Upper Surface of Wings: Forewings varying from grayish white or pale yellowish brown to pale ochraceous salmon or brown, either unicolorous or with basal, median, and terminal areas pale, having broad brown or dark brown bands basad of t. a. and distad of t. p. lines; cross lines usually present, dark brown, obsolescent or absent in some specimens; t. a. line arising on costa one-third distance from base, outwardly angled below radial vein and in cubital cell, angled basad to meet inner margin about three-tenths of distance from base; discal spot blackish brown, prominent; median line absent; t. p. line arising on costa three-fourths distance from base, weakly concave to cell M1, with inward teeth on veins, outwardly bowed, then broadly concave, thickened on veins, meeting inner margin three-fourths distance from base; s t. line varying from broad, diffuse, partially shaded by dark brown, to obsolescent or absent; terminal line absent; fringe either concolorous with wing or with basal portion more or less darkened opposite veins. Hind wings pale grayish white, more or less evenly covered with widely spaced pale grayish brown to grayish brown scales; without macalation except for grayish brown discal dot; terminal line absent; fringe concolorous with wing" (Rindge 1975).

Fig. 3. Adult *male Erannis defoliaria* with wings extended (left) and folded (right)
[Images from: (left) http://www.habitas.org.uk/moths/set.asp?item=6026smup; (right) http://www.nic.funet.fi/index/Tree_of_life/insecta/lepidoptera/ditrysia/geometroidea/geometridae/ennominae/erannis/defoliaria-4m.jpg]

"Under Surface of [Male] Wings: Forewings grayish white to pale brown, with variable amount of dark gray and grayish brown scales; maculation varying from obsolescent, having discal dot and faint trace of t. p. line, to similar to that of upper surface but more weakly represented. Hind wings pale grayish white, evenly covered with grayish brown or dark brown scales; maculation similar to that of upper surface" (Rindge 1975).

"Length of [Male] Forewing: 17 to 25 mm [0.67-1 in]" (Rindge 1975).

Female. Head with vertex white; front and palpi brownish black. Thorax above white, with paired black spots on each segment; below brownish black~ legs black or brownish black and white. Abdomen above and below white, variably spotted with black, more or less grouped on dorsal surface to form two irregular longitudinal bands. Wing pads minute, black anteriorly, white posteriorly" (Rindge 1975).

Fig. 4. Lateral (left) and dorsal (right) view of adult female *Erannis defoliaria*
[Image from: http://www.mothsplus.co.uk/PICTURE%20PAGES/Mottled%20Umber%20female.htm]

Biology and Ecology

In Europe, *E. defoliaria* has one generation per year and overwinters in the egg stage (Browne 1968, Szöcs et al. 1993, reviewed in Ciesla 2003, Tikkanen et al. 2006). Development of *E. defoliaria* begins at approximately 5°C [41°F] (Reiff 1953, Tikkanen et al. 2006).

Adults are adapted to low temperatures, being present from October to December (Browne 1968, Skinner 1984, Warrington 1985, Szöcs et al. 1993). Adult females are "wingless" (apterous) and do not fly, but crawl to the host plant for mating and oviposition (Cao and Luciano 2004, Tikkanen et al. 2006). The female to male sex ratio is 10:4, and males mate several times (Kipiani et al. 1982). Females find their mates by emitting pheromones (reviewed in Hansson et al. 1990, Szöcs et al. 1993). Oviposition lasts approximately 15-25 days, and egg production decreases with decreasing temperatures (Kipiani et al. 1982). Eggs are laid singly or in groups on or near the host plant in sheltered areas including stems, bark crevices, or in lichens and moss (Browne 1968, reviewed in Ciesla 2003, Tikkanen et al. 2006). A single female may lay 250-400 eggs (MAFF 1978, Kipiani et al. 1982). Larvae hatch in spring and feed on buds, foliage and flowers, either in the open or concealed within leaves bound together with silk (Browne 1968, MAFF 1978). Larvae are present throughout spring (April-late June); there are 5 instars (Warrington 1985). In Italy, larval densities ranged from 1-18 larvae per 40 branch tips (Cao and Luciano 2004). Pupation occurs in summer in soil near the base of the host plant and adults emerge in the fall (Chrystal 1937, Browne 1968, Porter 1997, reviewed in Ciesla 2003).

See 'Known Hosts' (below) for a listing of the plants that can be attacked by *E. defoliaria* and 'Pest Importance' for a discussion of the potential economic impact.

Pest Importance

The economic impact of *E. defoliaria* is difficult to measure especially because this pest typically occurs with other primary defoliators (including *Operophtera brumata* and *Tortrix viridana*) and secondary pests (Browne 1968). Additionally, considerable defoliation can occur without causing significant damage or tree death (Mattson and Addy 1975, Furniss and Carolin 1977, Drooz 1985). Heavy defoliation repeated over a period years can cause decline of tree health, reduced growth rates, attraction of secondary pests, and tree death (Furniss and Carolin 1977, Drooz 1985).

Erannis defoliaria is considered a pest of economic importance in northern and middle Europe, however reports of outbreaks are rare (reviewed in Hansson et al. 1990, Humphrey and Swaine 1997). Heavy infestations by polyphagous defoliators including *E. defoliaria* have reportedly caused damage to oak forests in northeast Romania (Ciornei and Mihalache 1998). Occasionally this pest causes significant damage in European orchards (reviewed in Hansson et al. 1990).

Risks associated with *E. defoliaria* have been evaluated for North American forests. The insect was considered of very high risk, and this rating was very certain (Ciesla 2003). Establishment, spread, and environmental-impact potentials were considered high, while economic-impact potential was moderate.

Symptoms

Feeding damage by *E. defoliaria* larvae is similar to that caused by other polyphagous defoliators. Evidence of the pest includes pupal cases in the soil near the base of host trees; rolled leaves of host trees spun with silk webbing; feeding damage on buds, foliage and flowers of host trees.

Known Hosts

E. defoliaria attacks a wide range of deciduous tree species:

Hosts	References
Acer sp. (maple)	(Ciesla 2003, CAB 2006)
Acer campestre (field maple)	(CAB 2006)
Acer pseudoplatanus (sycamore)	(CAB 2006)
Amelanchier sp. (serviceberry)	(Ciesla 2003)
Betula sp. (birch)	(Browne 1968, Zhang 1994)
Corylus sp. (hazelnut)	(Zhang 1994)
Corylus avellana (filbert, hazelnut)	(Ciesla 2003)
Cotoneaster sp. (cotoneaster, hollyberry)	(Zhang 1994, Ciesla 2003)

Hosts	**References**
Crataegus sp. (hawthorn)	(Hansson et al. 1990, Zhang 1994)
Crataegus oxyacantha (hawthorn, thornapple)	(Ciesla 2003)
Fagus sp. (beech)	(Browne 1968, Zhang 1994)
Fagus sylvatica [= "*silvatica*"] (European beech)	(Ciesla 2003, CAB 2006)
Hippophae rhamnoides (seaberry)	(Ciesla 2003)
Tilia sp. (linden)	(Ciesla 2003)
Tilia platyphylla (largeleaf linden)	(Ciesla 2003)
Lonicera sp. (honeysuckle)	(Zhang 1994)
Lonicera xylosteum (dwarf honeysuckle)	(Ciesla 2003)
Malus sp. (apple)	(Zhang 1994)
Malus domestica (apple)	(Ciesla 2003)
Nothofagus alpina (beech)	(CAB 2006)
Nothofagus procera (beech)	(Welch and Greatorex-Davies 1993)
Prunus sp. (cherry, peach, plum)	(Hansson et al. 1990, Zhang 1994, Ciesla 2003)
Prunus padus (European bird cherry)	(Ciesla 2003)
Prunus spinosa (blackthorn)	(Ciesla 2003)
Pyrus sp. (pear)	(Zhang 1994)
Quercus sp. (oak)	(Browne 1968, Zhang 1994)
Quercus petraea (durmast, sessile oak)	(Ciesla 2003, CAB 2006)
Quercus pubescens (downy oak)	(Cao and Luciano 2004)
Quercus robur (common oak)	(Ciesla 2003, CAB 2006)
Quercus rubra (northern red oak)	(CAB 2006)
Ribes sp. (currant, gooseberry)	(Zhang 1994)
Ribes alpinum (currant)	(Ciesla 2003)
Ribes nigrum (currant)	(Ciesla 2003)
Ribes rubrum (currant)	(Ciesla 2003)
Rosa sp. (rose)	(Zhang 1994, Ciesla 2003)
Sorbus aucuparia (European mountain ash)	(Ciesla 2003)
Sorbus hybrida (oakleaf mountain ash)	(Ciesla 2003)
Sorbus intermedia (Swedish mountain ash)	(Ciesla 2003)
Ulmus sp. (elm)	(Browne 1968, Ciesla 2003)

Known Distribution

Erannis defoliaria is widely distributed throughout Europe and was introduced and established on the North American Pacific coast, including British Columbia (Browne 1968).

Location	**References**
Austria	(CAB 2006)
Belgium	(CAB 2006)
Bulgaria	(CAB 2006)
Czech Republic (former Czechoslovakia)	(CAB 2006)
Finland	(Ciesla 2003)
France	(CAB 2006)
Georgia, Republic of	(Ciesla 2003)
Germany	(CAB 2006)
Hungary	(Hansson et al. 1990, Szöcs et al. 1993, CAB 2006)
Italy	(Cao and Luciano 2004)
Japan	(Zhang 1994)
Netherlands	(CAB 2006)
North America[1] (Pacific coast, British Columbia, Canada)	(Browne 1968, Rindge 1975, Ciesla 2003)
Norway	(Zhang 1994, CAB 2006)
Poland	(CAB 2006)
Romania	(Ciornei and Mihalache 1998)
Russian Federation (Former USSR)	(Zhang 1994, CAB 2006)
Scotland	(Tikkanen et al. 2006)
Spain	(Zhang 1994, CAB 2006)
Sweden	(Ciesla 2003)
Switzerland	(CAB 2006)
United Kingdom (British Isles)	(Browne 1968, Zhang 1994, CAB 2006)

1. Possibly confused with *Erannis tiliaria*.

Potential Distribution within the US

Based on the reported distribution of the species outside North America, *E. defoliaria* is most likely to be associated with biomes classified as temperate-broadleaf-and-mixed forest. This biome accounts for 28% of the area within the contiguous US and is most common in the eastern US. A small pocket occurs in the Pacific Northwest. However, this biome does not occur in British Columbia (see 'Known Distribution'). British Columbia has temperate grasslands, savannas and shrublands, temperate coniferous forests; and boreal forests. The distribution of *E. defoliaria* in British Columbia has not been described, so there is a chance the insect can survive in all of these biomes. However, the insect is more likely to be associated with temperate coniferous forests, given the types of biomes that occur within its native range. Temperate coniferous forests account for an additional 19% of the area of the contiguous US and scattered through the Pacific Northwest, the Intermountain West, and the Southeast.

Survey

Because larvae tend to be in the upper foliage of trees, visual inspection of leaves is not recommended for survey. To inspect for larvae, beat sample or pruning samples should be collected. For beat samples, relatively low peripheral branches, at a height of approximately 6 ft [2 m], should be shaken vigorously over 0.8 m^2 trays to dislodge any larvae; 25 such sample units should be collected from a site (Welch and Greatorex-Davies 1993). For pruning samples, the terminal 20 inches [50 cm] should be clipped using a pole pruner. Branches should be 20-30 feet [6-9 m] off the ground (Welch and Greatorex-Davies 1993). It is helpful to affix a frame with a plastic bag to the pole pruner to catch branch tips as they are cut. Five such branches should be cut from a site (Welch and Greatorex-Davies 1993).

Pheromones are available to detect adult male *E. defoliaria* (El-Sayed 2004). The compounds (Z,Z,Z)-3,6,9-nonadecatriene and (Z,Z)-3,9-*cis*-6,7-epoxy-nonadecadiene were extracted in a 1:3 ratio from the abdomens of females (Hansson et al. 1990). A 1:3 mixture of the compounds also attracted male *Colotois pennaria* and *Agriopis marginaria* (Hansson et al. 1990). The enantiomer (Z,Z)-3,9-*cis*-6S,7R-epoxy-nonadecadiene was far more attractive than the 6R,7S enantiomer, which did not attract any *E. defoliaria* (Szöcs et al. 1993). The attractive enatiomer was most effective when combined in a 1:1 ratio with (Z,Z,Z)-3,6,9-nonadecatriene (Szöcs et al. 1993). This mixture will also attract male *E. tiliaria tiliaria* and *E. tiliaria vancouverensis* (Szöcs et al. 2001). The lure should be used in conjunction with Pherocon 1C traps or triangular traps made from plastic sheets with at least one 4x6 inch [10x16 cm] surface covered with tanglefoot (Szöcs et al. 2001). Traps should be placed abut 5 ft [1.5 m] above the ground and should be separated from each other by at least 330 ft [100 m] (Szöcs et al. 2001).

Surveys for *E. defoliaria* will be complicated because the insect closely resembles *Agriopis marginaria* (a.k.a. dotted border) and *E. tiliaria* (linden looper) (Rindge 1975, Furniss and Carolin 1977, Porter 1997, Szöcs et al. 2001, Beccaloni et al. 2003, FUNET 2006). *Agriopis marginaria* occurs in Europe, while *E. tiliaria* is a common North American species (Rindge 1975, Szöcs et al. 2001). All three species share a similar host range including *Quercus* spp. (Furniss and Carolin 1977, Porter 1997, FUNET 2006). *Agriopis marginaria* is distinguished by a row of dots on the hindwing margin (Skinner 1984). Rindge (1975) notes that *E. defoliaria* and *E. tiliaria* look very much alike and have only extremely subtle differences in maculation and genitalia. In the laboratory, *E. defoliaria* will mate with *E. tilaria* and produce offspring, but the fertility of these offspring is unknown (Rindge 1975). Consequently, North American

Erannis are treated as a single species *E. tiliaria*, consisting of two subspecies (*E. tiliaria tiliaria* and *E. tiliaria vancouverensis*), and *E. defoliaria* remains a distinct species not currently known to occur in the US (Rindge 1975).

Fig. 5. Forms of *Erannis tiliaria*, common in North America: (upper left) larva, (upper right, lower left) adult male, (lower right) adult female.
[Images from (upper left) L.R. Barber http://www.forestryimages.org/browse/detail.cfm?imgnum=0795033; (all others) http://www.cbif.gc.ca/spp_pages/geometroidea/phps/geo2_e.php]

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Leucoma salicis

Fig. 1. Drawing of *Leucoma salicis* larva
[Image from http://www.whatsthiscaterpillar.co.uk/plates.htm]

Scientific Name

Leucoma salicis Linnaeus

Synonyms:
Bombyx salicis Linnaeus
Laelia salicis Linnaeus
Liparis salicis Linnaeus
Phalaena salicis Linnaeus
Stilpnotia salicis Linnaeus

Common Name

satin moth, white satin moth

Type of Pest

Moth, defoliator

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera, **Family:** Lymantriidae, **Subfamily:** Orgyiinae

Reason for inclusion in manual

Exotic Forest Pest Information System – classified as a moderate risk pest with the potential to attack oaks

Pest Description

“The wings of both male and female moths are pure white with a satin like luster. Wing expanse is from 1½ to 2 inches. The head, thorax, and abdomen are black, but are so densely covered with long white hairs that the body appears white unless these hairs have been removed” (Brown 1940).

“Males 15 to 20 mm, females 20-25 mm in length” (Glendenning 1924).

“[*Leucoma salicis*] is the only moth of its size in [the US] fauna that is entirely white, including the body, and with each leg marked by a series of annular black bands or at least transverse black bars (which may not always form complete rings). Some of the white species of Arctiidae may nearly fit this description but always have a patch of color somewhere on the body, spotted wings, or legs without black markings. The body integument of [*L. salicis*], wherever the hairy vestiture has been worn thin or rubbed off, may be seen to be very dark, almost black. The antennae of both sexes are clearly of the lymantriid type, with usually one long and one very short spinule arising from the tip of each branch. The wings, especially the forewings, have a satinlike lustre which gave rise to the common name. Females especially may vary considerably in size, like those of the gypsy moth, but otherwise hardly any variation is apparent ” (Ferguson 1978).

“The eggs are laid in small oval-shaped patches about five-eighths inch long and one-half inch wide which are covered with a glistening-white secretion” (Brown 1940).

“Eggs spherical, but slightly flattened; light green when freshly laid, gradually becoming brown as embryo develops” (Glendenning 1924).

Fig. 2. Life stages of Leucoma salicis: eggs (upper left), larva (upper right), adult male (bottom).
[Images (above) from Gyorgy Csoka, www.forestryimages.org; (bottom) Swedish Natural History Museum. http://swedish.naturalhistory.museum/en/svenska_fjarilar/l/images/leucoma_salicis_male.gif]

“The full-grown larva is about 1-3/8 inches long. The head is black with a bluish tinge. The body is black on the upper side with a row of large white blotches down the middle. The sides of the body are mottled with black and white, and each segment bears a transverse row of reddish-brown tubercles from which arise yellowish-brown hairs" (Brown 1940).

"The somewhat rounded dorsal spots lie astride the intersegmental membranes, and each tends to be bilaterally constricted near the middle at the intersegmental position. The verrucae (tubercles) are quite large, reddish brown, and bear relatively uniform tufts of brown to pale-yellowish hair; there are no dense tufts or hair pencils. The subdorsal verrucae are largest, the supraspiracular ones moderately large, and the sublateral ones smaller and double. The usual dorsal glands are present on abdominal segments six and seven. The head is black and sparsely clothed in rather long hairs. The very conspicuous whitish dorsal spots serve to distinguish this species from all other Lymantriidae in North America" (Ferguson 1978).

Fig. 3. Adult *Leucoma salicis* in repose. Note banding pattern on legs.
[Image from Lynette Schimming, bugguide.net/node/view/27838]

"The pupa, which is about 7/8 inch long, is shining black and covered with long white and golden hairs. The cocoon is loosely constructed, and the pupa within is clearly visible" (Brown 1940).

Biology and Ecology

Leucoma salicis is native to Asia and Europe and was first detected in North America in 1920 (Burgess and Crossman 1927). This insect has one generation per year in North America and most of Europe, but two generations per year have been reported in southern Europe (Rosovsky 2001). Moths emerge over a period of 3-4 weeks beginning in late June. Mating occurs a few hours following eclosion. Females may mate more than once, particularly if a disturbance interrupts oviposition. Pairs may be observed mating for several hours. During heavy infestations, mating pairs may be observed on trees, brush grass and non-living structures such as utility poles and light posts (Burgess and Crossman 1927). Older literature suggests moths are strong fliers (Burgess and Crossman 1927, Brown 1940), and are attracted to light (Burgess and Crossman 1927). In

Europe, *L. salicis* has been reported to migrate in large groups (Serafimovski 1954). However, Browne (1968) explains that while males are active, females are "sluggish and rarely fly." Moths are nocturnal, but will fly if disturbed during the day. Adult *L. salicis* live approximately 7-14 days (Burgess and Crossman 1927, Serafimovski 1954, Wagner and Leonard 1979a, reviewed in Barbosa and Schaefer 1997, reviewed in Rosovsky 2001).

Oviposition occurs in July. Females deposit between 316-516 eggs in oval patches covered in a satin-white secretion. Eggs are primarily deposited on stems and lower crowns of hosts, but may be placed on inanimate structures. (Reeks and Smith 1956). Under laboratory conditions, 500-1000 were produced (571 on average), and hatch occurred in about 14-15 days (Burgess and Crossman 1927, Reeks and Smith 1956, reviewed in Barbosa and Schaefer 1997).

Larvae develop through 6 or 7 instars (Burgess and Crossman 1927, Serafimovski 1954). Neonates begin feeing soon after hatch. Because this insect overwinters as a third instar larva, third through seventh instars occur from April through June while first through third instars occur from mid July to late September (part of the next generation). Young larvae may be able to disperse like other lymantriids by dropping on a trailing silk thread and utilizing air and wind currents to "balloon" to other locations (Barbosa and Schaefer 1997, Zlotina et al. 1999). In preparation for overwintering, second instars stop feeding and build a hibernaculum from strong webbing. Hibernacula are often in bark cracks or other depressions and can be difficult to see (Burgess and Crossman 1927). A second instar enters the hibernaculum, molts, and prepares to overwinter. Typically, one larva occurs within a hibernaculum, but in heavy infestations, more than one larvae may be present (Burgess and Crossman 1927). First through third instars feed on the surface of leaves, but third instars typically do not eat until spring (Burgess and Crossman 1927). Later instars will consume the entire leaf except for large veins (Burgess and Crossman 1927). Fourth through sixth instars molt in bark crevices covered with webbing. Sixth or seventh instars pupate inside loosely spun cocoons on slightly rolled leaves, in bark crevices or in other sheltered areas near host trees. Adults emerge about ten days later (Burgess and Crossman 1927, reviewed in Barbosa and Schaefer 1997).

When population density is high, larval mortality increases (from factors including parasitism by Hymenoptera and Diptera, infection by entomopathogenic fungi, and predation by insects and vertebrates) and helps reduce the duration of outbreaks (Wagner and Leonard 1980, Ziemnicka 1981, Ziemnicka and Sosnowska 1996, Zurek and Keddie 2000).

Pest Importance

Leucoma salicis is considered a periodic pest of economic importance, defoliating *Populus* spp. and *Salix* spp. since its introduction to North America (Burgess and Crossman 1927, Brown 1940, Wagner and Leonard 1979b, Ziemnicka 1981, Rosovsky 2001, CAB 2006). This pest has also been reported as a pest of concern in Turkey (Çobanoğlu 1992).

Fig. 4. Severe defoliation of *Populus* sp. caused by *Leucoma salicis*.
[Image from Canadian Ministry of Forests and Range,
http://www.for.gov.bc.ca/hfp/publications/00198/301%20to%20350/fig333.jpg]

Leucoma salicis can be a nuisance pest. Burgess and Crossman (1927) report instances of *L. salicis* larvae dropping out of trees onto pedestrians, crawling into homes, and swarming over fences, buildings, and sidewalks. Serafimovski (1954) claims that in addition to damaging trees, this moth also "makes the streets dirty by its excrements." In North America the satin moth often occurs with other defoliators, such as tent caterpillars (i.e., *Malacosoma* spp.). Satin moths can completely defoliate trees, and repeated severe defoliation can result in reduced radial growth, top-kill and tree mortality. Ninety percent of feeding occurs in the spring (Barbosa and Schaefer 1997).

This moth was first discovered in the United States and Canada in 1920 (Brown 1940, Wagner and Leonard 1980, Drooz 1985, Barbosa and Schaefer 1997, Rosovsky 2001, reviewed in CAB 2006). The species was introduced at least twice: once in Boston, Massachusetts and once in New Westminster, British Columbia (Burgess and Crossman 1927, Ferguson 1978). Defoliation of *Populus* spp. by *L. salicis* was first noted in 1920 (Burgess and Crossman 1927). The area infested by *L. salicis* in North America has increased linearly since its introduction, and its rate of spread has been faster than that of gypsy moth (Barbosa and Schaefer 1997). However, Barbosa and Schaefer (1997) note that while poplars were commonly planted as shade, street, and park trees around the time *L. salicis* was introduced, they are "currently rare in the same area today". Reeks and Smith (1956) also observed the decline in popularity of shade poplars in Canada during the early part of the moth's spread across North America.

The infested trees will refoliate after being stripped of leaves by *L. salicis* and can tolerate repeated defoliations (Reeks and Smith 1956, Drooz 1985, Rosovsky 2001). Defoliation can result in decline in overall growth and development, attack by secondary pests (Drooz 1985). Multiple defoliations can lead to death of branches. In severe infestations, the tree can be killed outright (Reeks and Smith 1956, Ziemnicka and Sosnowska 1996).

Rosovsky (2001) suggested that *L. salicis* poses a moderate risk to North American forests, and this assessment was very certain (Rosovsky 2001). Because the insect is already present on the continent, risks stemmed from the potential of *L. salicis* to continue to spread into new areas. Establishment potential was rated high, largely because the insect has already established in North America. The potential for spread, economic damage, and environmental harm were considered moderate, respectively. Preferred hosts, *Populus* spp., are of limited economic value and can recover quickly from damage (Rosovsky 2001).

Symptoms

Damage by *L. salicis* is similar to that caused by larvae of other skeletonizing moths and sawflies. *Leucoma salicis* larvae are gregarious defoliators, able to consume whole leaves and sometimes avoid tough veins in older foliage growth. Eggs masses, larvae and pupae may also be evident on trees and other inanimate structures (Drooz 1985). Frass droppings may be evident under hosts if population density is high (Serafimovski 1954).

Known Hosts

Leucoma salicis attacks a wide range of deciduous tree species. It is named for its preferred hosts within the family Salicaceae (Wallner 1988, Barbosa and Schaefer 1997, reviewed in Rosovsky 2001). Burgess (1927) describes scrub oak (*Quercus ilicifolia*) and black oak (*Q. velutina*) as partially favorable, and red oak (*Q. rubra*), white oak (*Q. alba*), and scarlet oak (*Q. coccinea*) as non-favorable.

Hosts	**References**
Acer sp. (maple)	(Sun 1988, Rosovsky 2001)
Alnus sp. (alder)	(CAB 2006)
Betula sp. (birch)	(CAB 2006)
Corylus sp. (hazelnut)	(Sun 1988)
Malus sp. (crabapple)	(CAB 2006)
Populus sp. (poplar)	(Serafimovski 1954, Reeks and Smith 1956, Ziemnicka 1981, Grijpma et al. 1987, Grijpma 1988, Avtzis 1990, Humphreys 1996, Barbosa and Schaefer 1997, Gries et al. 1997, Allegro 1999a, Zurek and Keddie 2000, Rosovsky 2001, Szöcs et al. 2005, CAB 2006)

Hosts	**References**
Populus alba (white poplar)	(Brown 1940, Reeks and Smith 1956, Wagner and Leonard 1979b, Grijpma 1988, Avtzis 1990)
Populus alba var. *pyramidalis* (Boll's poplar)	(CAB 2006)
P. balsamifera (="*Populus balsamica*"?) (black cottonwood)	(Ziemnicka and Sosnowska 1996)
Populus balsamifera ssp. *balsamifera* (=*P. candicans*) (balm-of-Gilead)	(Brown 1940, Reeks and Smith 1956, Wagner and Leonard 1979b, Barbosa and Schaefer 1997)
Populus canadensis (hybrid black poplar)	(Serafimovski 1954, CAB 2006)
Populus ×canescens (=*P. alba × tremula*) (gray poplar)	(Grijpma 1988, Çobanoğlu 1992)
Populus deltoides (eastern cottonwood) [1]	(Brown 1940, Wagner and Leonard 1979a, Grijpma 1988, Wallner 1988, Barbosa and Schaefer 1997, Allegro 1999a, Rosovsky 2001)
Populus eugenei (Carolina poplar)	(Reeks and Smith 1956)
Populus grandidentata (bigtooth aspen)	(Brown 1940, Wagner and Leonard 1979a, 1980, Barbosa and Schaefer 1997, CAB 2006)
Populus nigra (black poplar)	(Serafimovski 1954, Grijpma 1988, Ziemnicka and Sosnowska 1996, Allegro 1999a, CAB 2006)
Populus nigra var. *italica* (=*P. pyramidalis*) (Lombardy poplar)[1]	(Brown 1940, Serafimovski 1954, Reeks and Smith 1956, Wagner and Leonard 1979a, Ziemnicka and Sosnowska 1996, Barbosa and Schaefer 1997, Rosovsky 2001)
Populus simonii (Simon's poplar)	(Serafimovski 1954, Wagner and Leonard 1979a)
Populus tremula (European aspen)	(Grijpma 1988)
Populus tremuloides (trembling aspen)	(Brown 1940, Reeks and Smith 1956) (Wagner and Leonard 1979a, 1980, Grijpma 1988, Humphreys 1996, Barbosa and Schaefer 1997, Allegro 1999a, CAB 2006)
Populus trichocarpa (black cottonwood)[2]	(Brown 1940, Grijpma 1988, Humphreys 1996, Allegro 1999a)
Prunus sp. (stone fruit)	(CAB 2006)
Quercus ilicifolia (scrub oak)	(Burgess and Crossman 1927)
Quercus velutina (black oak)	(Burgess and Crossman 1927)

Hosts	**References**
Salix sp. (willow)	(Serafimovski 1954, Reeks and Smith 1956, Ziemnicka 1981, Grijpma et al. 1987, Grijpma 1988, Sun 1988, Ziemnicka and Sosnowska 1996, Barbosa and Schaefer 1997, Gries et al. 1997, Zurek and Keddie 2000, Rosovsky 2001, Szöcs et al. 2005, CAB 2006)
Salix alba (white willow)	(CAB 2006)
Salix alba var. *vitellina* (golden willow)	(Brown 1940)
Salix babylonica (weeping willow)	(Wagner and Leonard 1979a)
Salix caprea (pussy willow)	(CAB 2006)
Salix cinerea (large gray willow)	(Wistow 1992)
Salix lasiandra (=*S. lucida* ssp. *lasiandra*) (Pacific willow)	(Brown 1940)
Salix scouleriana (Scouler's willow)	(Brown 1940)

1. Listed in Wallner (1988) and reviewed in Rosovsky (2001) as a "preferred" host, and in Wagner and Leonard (1979b) as "most suitable" hosts.
2. Grijpma (1988) reports 100% mortality for larvae reared on leaves of *P. trichocarpa*. The larvae demonstrated "symptoms of hormonal deviations."

Known Distribution

Leucoma salicis is reported from:

Location	**References**
Austria	(CAB 2006)
Belgium	(CAB 2006)
Bulgaria	(CAB 2006)
Canada	(Brown 1940, Reeks and Smith 1956, Wagner and Leonard 1980, Wallner 1988, Humphreys 1996, Barbosa and Schaefer 1997, Gries et al. 1997, Zurek and Keddie 2000, Szöcs et al. 2005, CAB 2006)
China	(Sun 1988, Avtzis 1990, Rosovsky 2001, CAB 2006)
Czechoslovakia (former)	(CAB 2006)
France	(CAB 2006)
Germany	(CAB 2006)
Greece	(Avtzis 1990, CAB 2006)
Hungary	(Serafimovski 1954, Szöcs et al. 2005, CAB 2006)
Ireland	(CAB 2006)
Italy	(Serafimovski 1954, Allegro 1999b, 1999a, CAB 2006)
Japan	(Rosovsky 2001, CAB 2006)

Location	References
Korea	(Rosovsky 2001, CAB 2006)
Macedonia	(CAB 2006)
Netherlands	(CAB 2006)
Poland	(Ziemnicka 1981, Ziemnicka and Sosnowska 1996, CAB 2006)
Romania	(CAB 2006)
Russia	(Rosovsky 2001, CAB 2006)
Serbia	(Serafimovski 1954, CAB 2006)
Spain	(CAB 2006)
Sweden	(CAB 2006)
Switzerland	(CAB 2006)
Turkey	(Çobanoğlu 1992, CAB 2006)
United Kingdom	(Wistow 1992, Rosovsky 2001, CAB 2006)
United States	(Brown 1940, Reeks and Smith 1956, Wagner and Leonard 1980, Wallner 1988, Humphreys 1996, Barbosa and Schaefer 1997, Rosovsky 2001, CAB 2006)
Yugoslavia	(Serafimovski 1954, CAB 2006)

Potential Distribution within the US

Based strictly on the reported distribution of *L. salicis* outside the United States, we predict that this insect is most closely affiliated with temperate broadleaf and mixed forest. However, this prediction is based on an extremely coarse description of the distribution (country level), and does not account for the exact distribution of the species within each country. On the Pacific coast of North America, *L. salicis* has also established in an area generally classified as temperate coniferous forest. Thus, both forested biomes seem suitable for the species. These biomes account for 47% of the contiguous US and are most common in the eastern US, Pacific Northwest, and higher elevations of the Intermountain West.

Survey

Until recently, visual inspections of trees were the only recommended option to detect infestations of *L. salicis*. Tree trunks were examined for the presence of overwintering larvae in hibernacula (reviewed in CAB 2006) and foliage was inspected for feeding damage and the insect itself (Ostry et al. 1988).

Pheromone lures for *L. salicis* recently became available. The major sex pheromone was identified as (3*Z*)-*cis*-6,7-*cis*-9,10-diepoxy-3-henicosene and named “leucomalure” (Gries et al. 1997). Szöcs et al. (2005) later suggested that the isomer (3*Z*,6*R*,7*S*,9*R*,10*S*)-*cis*-6,7-9,10-diepoxy-3-henicosene was the primary sex pheromone. This compound on its own attracted significant numbers of male *L. salicis* (Szöcs et al. 2005). Leucomalure has proven attractive to North American populations, but not to European populations. Conversely, (3*Z*,6*R*,7*S*,9*R*,10*S*)-*cis*-6,7-9,10-diepoxy-3-henicosene was attracted *L. salicis* in Europe but not in North America. Szöcs et al. (2005) speculate that the North American population may have come from Asia, not Europe. Pheromone-baited delta traps have been used effectively to detect populations of *L. salicis* (Gries et al. 1997, Szöcs et al. 2005). Lures should be prepared by placing approximately 50μg of the compound in 20μl of hexane onto rubber tubing or rubber septa (Szöcs et al. 2005). Pheromone-baited traps should be placed 1.5 m [5 ft] above ground surface and traps should be a minimum of 15 m [50 ft] apart (Gries et al. 1997).

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Lymantria dispar (Asian strain)

Scientific Name

Lymantria dispar Linnaeus

<u>Synonyms:</u>
Bombyx dispar
Hypogymna dispar
Liparis dispar
Ocneria dispar
Phalaena dispar
Phaloena dispar
Porthesia dispar
Porthetria dispar

Common Names

Asian gypsy moth

Type of Pest

Moth, defoliator

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera,
Family: Lymantriidae

Reason for inclusion in manual

Exotic Forest Pest Information System – classified as a high risk pest with the potential to attack oaks

Pest Description

Female flight is a key feature that distinguishes Asian from European gypsy moths. Females of European gypsy moth cannot fly, but females of Asian gypsy moth can (see ‘Biology and Ecology’). Asian and European gypsy moths appear nearly identical and cannot be distinguished reliably in the field unless females are observed in flight (Wallner 2000).

European *L. dispar* have the following form: “Adult male, antennae pectinate, light brown; body slender, dark brown; wings light to dark brown with black markings: blackish, wavy bands on forewings with arrowhead markings near anal edge, wingspan 2.5-3.8 cm. Adult female, antennae pectinate, but narrower than male; body stout, cylindrical with yellow hairs; wings white or grayish-white with black markings: brown or blackish bands and arrowhead markings on forewings, a marginal transverse line of dark-colored dots on outer edge of fore and hind wings. Egg globular, small (1.10-1.25 mm diameter), and white or transparent. Egg mass or clump oval, raised in center, covered with buff-colored hairs (from female's abdomen and sides), appearance similar to chamois cloth or sponge, 1.3-5.1 cm long. Mature larva length 37-60 mm [~1 ½-2 ½ in]; head with yellow markings; body with dusky or slate colors, peppered with numerous small dark spots, 3 light stripes along back, 5 pairs of blue spots followed by 6 pairs of brick-red spots on segments 2-12, general ventral surface light grayish-brown; 1st instar ca. 3 mm long, extremely hairy and dark. Pupa reddish-brown; few short light brown or red hairs around spiracles and across thoracic and abdominal segments; length 1.9-2.5 cm; loosely attached to objects with a few strands of silk" (Fig. 1) (Dixon and Foltz 1985).

Fig. 1. Life stages of Asian gypsy moth: (upper left) adult female; (upper right) adult male; (lower left) egg masses and neonate larvae; and, (lower right) larva. Images not to scale.
[All images from J. Ghent, www.forestryimages.org]

Coloration of larvae from Asian populations of *L. dispar* varies considerably from yellow to "black-backed" to "mottled dark gray," the most common color (Sun 1988).

Biology and Ecology

The biology and ecology of both the Asian and European forms of *L. dispar* are similar. The primary differences are: (i) Asian female moths fly (>20 km [>12 mi]) while European female gypsy moths are flightless; and (ii) the Asian strain has slightly different host preferences than the European strain (reviewed in Drooz 1985, Reineke and Zebitz 1998, Charlton et al. 1999, reviewed in Wallner 2000). Asian and European gypsy moths can hybridize under laboratory conditions, and traits of the hybrids (such as larval color, adult size, or female flight capacity), rarely resemble the European parents; hybrids either resemble the Asian parent or are intermediate between both parents (Garner and Slavicek 1996, Keena 1996, Keena et al. 2001).

Asian and European *L. dispar* have one generation per year (Drooz 1985, Sun 1988). In China, adults begin to emerge in late June and peak flight occurs in mid- to late July (Sun 1988). In the US, adults, descendents of European gypsy moth, appear in late July and August (Drooz 1985). Asian gypsy moths fly at dusk when light intensity reaches <3 *lux* for 2-3 hours, or when disturbed (Wallner et al. 1995, Charlton et al. 1999, reviewed in Keena et al. 2001). European male gypsy moths fly in a zigzag pattern approximately 1 m [~3 ft] above the ground (Drooz 1985). Flight of male Asian gypsy moths has been observed between 11-12°C [~52-55°F], while flight activity for the European form ceases below 17°C [63°F] (Cardé et al. 1996). In central Siberia, flight of females has been reported at temperatures as low as 11-13°C [~52-55°F] (Charlton et al. 1999).

After mating, females deposit between 100-1200 eggs in distinctive oval silky hair-covered masses on trunks, branches and other sheltered areas (i.e., on vehicles, signs, under rocks, roofs, etc.) (reviewed in Drooz 1985, Sun 1988, reviewed in Wallner 2000). Eggs are the overwintering stage for European and Asian *L. dispar* (Drooz 1985, Sun 1988). Eggs for both strains typically undergo diapause, but while 95% of European eggs diapause, only 75% of Asian eggs do (reviewed in Barlow et al. 2000).

In China, the larval stage lasts approximately 6 weeks. Larvae hatch from late April to early May (Sun 1988). Newly hatched *Lymantria* larvae remain on the hair-covered egg cases for a period before moving to buds and leaves to feed in the crown (Sun 1988). It is not known whether the larvae receive some nutritive benefit from the mass prior to feeding on foliage (Roonwal 1979). Males and females progress though five and six instars, respectively (Wallner 2000). *Lymantria dispar* larvae possess the ability to disperse by dropping on a trailing silk thread and utilizing air and wind currents to "balloon" to other locations (Zlotina et al. 1999). Late instar larvae exhibit a nocturnal feeding behavior and may be found during the day gathered in sheltered areas (Drooz 1985). Mature larvae pupate in bark crevices, branches or concealed in other sheltered areas (Sun 1988). Pupae are present 10-14 days and may be found attached to trees or other protected areas in groups (Drooz 1985).

Lymantria dispar has historically demonstrated food preferences, but these preferences may depend on available hosts and food supply [see 'Known Hosts'] (Drooz 1985, Glare et al. 1998). In general, European *L. dispar* prefer to feed on oaks (*Quercus* spp.) and other broadleaves while Asian *L. dispar* seem to prefer broadleaf trees and larch (*Larix* spp.) (reviewed in Wallner 2000). The selection of a location for egg deposition may also depend on the presence or density of other egg masses, host preference, and the extent of feeding that has already occurred on a host (Drooz 1985). In outbreak years, *L. dispar* tends to lay eggs on many tree species, including non-hosts, and other structures [see 'Survey'] (reviewed in Wallner 2000).

Population density is influenced by several factors including the available food supply, host selection and quality, and presence of natural enemies (Drooz 1985). When population density is high, parasitism by Hymenoptera, infection by polyhedral viral disease or introduced fungal pathogen, *Entomophaga maimaiga*, may result in high mortality of larvae and pupae (Pemberton et al. 1993, Glare et al. 1998, Solter 1999, Barlow et al. 2000, Ebling et al. 2004, Nielsen et al. 2005a, Nielsen et al. 2005b).

Pest Importance

Lymantria dispar causes significant defoliation of hardwood and fruit trees in Asia (Montgomery and Wallner 1988, Sun 1988, Wallner 2000), but damage is not constant. European and Asian gypsy moth populations undergo strong cycles. Outbreaks occur every 8-10 years in the northeastern US and China (Drooz 1985). An outbreak will typically last 1-3 years (Sun 1988, Wallner 2000). Defoliation can reduce growth, accelerate decline, increase susceptibility to secondary pests, or, in cases of severe, repeated defoliation, cause tree death (Drooz 1985).

Control of gypsy moth populations is costly. Since 1980, more than $35 million has been spent annually to control European gypsy moth in the US (Wallner 2000). Efforts to eradicate Asian gypsy moth from North America caused $25 million (reviewed in Wallner 2000).

Gypsy moth causes economic damage through its adverse impacts on forest productivity and aesthetics. In Pennsylvania, gypsy moth was responsible for the loss of $72 million worth of timber. Because of the extensive volumes of frass produced by larvae (Liebhold and Elkinton 1988a), *L. dispar* can be a severe nuisance and limit recreational opportunities (reviewed in Wallner 2000). In extreme cases, gypsy moth can lower property values (reviewed in Wallner 2000). Other costs are incurred as gypsy moth impacts ecosystem function, but the value of those functions can be difficult to quantify.

Commercial insecticides are available for control, but in many natural settings, complex terrain limits the feasibility of this option, especially over large areas. However, as has been observed with European *L. dispar*, formulations of endotoxin from *Bacillus thuringiensis kurstaki* (e.g, *Bt-k*) may be applied aerially to localized populations (Myers et al. 1998, Myers and Hosking 2002). *Bt* is generally considered host specific (Lacey and Siegel 2000), but unintended impacts on non-target species, typically other Lepidoptera, have been noted especially after repeated applications (Lacey and Siegel 2000, Boulton 2004). Biological control seems to be a more viable option (reviewed in Rosovsky 2001).

Previous experience with the European gypsy moth demonstrates that predators, parasitoids, and pathogens might be introduced. In previous years, generalist agents (e.g., *Compsilura concinata*) were introduced, often with significant impacts on non-target species (reviewed in Syrett 2002). Current protocols for the screening of agents limit the likelihood of these severe impacts to non-target species (reviewed in Hoddle and Syrett 2002).

Risks posed by Asian *L. dispar* for North American forests have been evaluated previously. In the Exotic Forest Pest Information System, *L. dispar* was considered a very high risk, and this assessment was given with a very high degree of certainty (reviewed in Wallner 2000). Previous experiences with European gypsy moth and beachhead populations of Asian gypsy moth in the US underscore the substantial chance of establishment, spread, economic damage, and environmental damage.

Symptoms

Lymantria dispar larvae are gregarious defoliators, able to consume whole leaves and sometimes avoid tough veins in older foliage growth. Eggs masses, larvae and pupae may also be evident on trees (Drooz 1985). Frass droppings may be evident under hosts if population density is high (Liebhold and Elkinton 1988a, 1988b).

Known Hosts

Lymantria dispar is polyphagous on over 500 species (Glare et al. 1998). For brevity, we primarily report host genera. Gypsy moth larvae feed preferentially on *Quercus* sp., *Populus* sp., *Tilia* sp., *Betula* sp., *Larix* sp., *Salix* sp., *Ulmus* spp., *Malus* sp., and *Liquidambar* sp (reviewed in Gottschalk 1988). Asian gypsy moths prefer hardwoods, fruit trees, and larches. They tend not to prefer *Fraxinus* sp., *Thuja* sp., *Robinia* sp., *Juniperus* sp., *Platanus* sp., *Liriodendron tulipifera*, and *Abies balsamea*.

Hosts	**References**
Acer sp. (maple)	(Zhang 1994, Wulf 1996)
Alnus incana spp. *rugosa* (speckled alder) [1]	(Glare et al. 1998)
Alnus sp. (alder)	(Wulf 1996)
Betula sp. (birch) [1]	(Gottschalk 1988, Zhang 1994, Wulf 1996, Glare et al. 1998, Wallner 2000)
Callistemon brachyandrus (prickly bottlebrush)[2]	(Matsuki et al. 2001)
Carpinus sp. (hornbeam)	(Wulf 1996)
Castanea sp. (chestnut)	(Wulf 1996)
Corymbia maculata (spotted gum)[2]	(Matsuki et al. 2001)
Crataegus sp. (hawthorn)	(Orozumbekov et al. 2003)
Diospyros sp. (persimmon)	(Zhang 1994, Wallner 2000)
Eucalyptus gunnii x *Eucalyptus dalrympleana* (=*E. irbyi*)[2]	(Matsuki et al. 2001)

Hosts	**References**
Eucalyptus gunnii (cider gum)[2]	(Matsuki et al. 2001)
Eucalyptus risdonii (Risdon peppermint gum) [2]	(Matsuki et al. 2001)
Eucalyptus urnigera (urn gum)[2]	(Matsuki et al. 2001)
Fagus sp. (beech)	(Wulf 1996)
Juglans sp. (walnut)	(Zhang 1994, Orozumbekov et al. 2003)
Larix sp. (larch)	(Gottschalk 1988, Zhang 1994, Wulf 1996, Wallner 2000)
Liquidambar sp. (sweetgum)	(Gottschalk 1988, Zhang 1994)
Malus sp. (apple) [1]	(Gottschalk 1988, Zhang 1994, Glare et al. 1998, Wallner 2000, Orozumbekov et al. 2003)
Picea pungens (blue spruce)	(Zhang 1994)
Pistacia sp. (pistachio)	(Zhang 1994, Orozumbekov et al. 2003)
Populus sp. (aspen, poplar) [1]	(Wulf 1996); (Zhang 1994, Glare et al. 1998, Wallner 2000, Orozumbekov et al. 2003)
Prunus sp. (apricot, cherry, peach, plum)	(Zhang 1994, Orozumbekov et al. 2003)
Quercus sp. (oak)[1]	(Gottschalk 1988, Zhang 1994, Wulf 1996, Glare et al. 1998, Wallner 2000)
Salix sp. (willow) [1]	(Gottschalk 1988, Wulf 1996, Glare et al. 1998, Wallner 2000)
Sassafras albidum (sassafras)	(Zhang 1994)
Shorea robusta (sal tree)	(Beeson 1941, Roonwal 1953, Roonwal et al. 1962)
Tilia sp. (basswood) [1]	(Gottschalk 1988, Wulf 1996, Glare et al. 1998, Wallner 2000)
Ulmus sp. (elm)	(Wulf 1996); (Wallner 2000)

1. Glare et al. (1998) cite these as the “preferred hosts” of gypsy moth.
2. Experimental. Matsuki et al. (2001) found these hosts support Asian gypsy moth equally well as its preferred hosts *Quercus pubescens* and *Q. robur*.

Known Distribution

The Asian strain of *L. dispar* occurs widely from 20º-60º N in temperate and boreal forests of the Palearctic (Montgomery and Wallner 1988, Sun 1988, Wallner 2000). Asian gypsy moth has also established in Europe (reviewed in Wallner 2000).

Since 1981, there have been multiple introductions of Asian gypsy moths into North American ports (Bogdanowicz et al. 1993, Bogdanowicz et al. 1997, Zlotina et al. 1999) including Vancouver, British Columbia, Canada; Wilmington, North Carolina; Portland, Oregon; and the Seattle-Tacoma area of Washington.

Detected populations were successfully eradicated (reviewed in Wallner 2000, reviewed in USDA 2006). Asian gypsy moth was also detected in Idaho in 2004 (Livingston 2004) and Texas in 2006 (reviewed in NAPPO 2006). In response to both cases, aggressive treatment campaigns were implemented.

Location	**References**
China	(Sun 1988, Reineke and Zebitz 1998, Reineke et al. 1999)
France[1]	(Cardé et al. 1996)
Germany[2]	(Reineke and Zebitz 1998, Reineke et al. 1999, Wallner 2000)
Japan	(Nielsen et al. 2005a, Nielsen et al. 2005b)
Kazakhstan	(Wulf 1996, Orozumbekov et al. 2003)
Kyrgyzstan	(Orozumbekov et al. 2003)
Russia	(Keena et al. 2001, Nielsen et al. 2005a)
United States[3]	(Bowen 1991, Bogdanowicz et al. 1993, Zlotina et al. 1999, Matsuki et al. 2001, Ebling et al. 2004)
United Kingdom[4]	(Zhang 1994)
Uzbekistan	(Orozumbekov et al. 2003)

1. W.E. Wallner, unpublished data (reviewed in Cardé et al. 1996).
2. Reineke et al. (1999) report the appearance of flying gypsy moth females in southwest Germany between 1992-1994. Reineke and Zebitz (1999) found female flight to be a differentiating characteristic between the European and Asian biotypes, but were unable to determine whether the southwest German gypsy moth females belonged to the Asian biotype.
3. There have been multiple introductions of Asian gypsy moth to North America; all introduced populations have been successfully eradicated.
4. Introduced and eradicated (“extinct”) (Zhang 1994)

Potential Distribution within the US

Based on the reported geographic distribution of Asian gypsy moth, it appears that the insect is most closely associated with biomes characterized as temperate coniferous forest, temperate broadleaf-and-mixed forest, and tropical and subtropical moist broadleaf forest. Uzbekistan reportedly does not have any of these three forested biomes, yet it has gypsy moth. Little is known about the distribution or behavior of gypsy moth in this region of Asia (Orozumbekov et al. 2003). More detailed information on the population dynamics here would be useful to predict the potential for gypsy moth to establish on trees in areas that are more generally classified as temperate grasslands and savannas.

Nevertheless, the three forested biomes certainly are at risk. All of these biomes are present and collectively account for approximately 47% of the area in the contiguous US. Detections of Asian gypsy moth in the US have generally occurred in areas with temperate coniferous forest or temperate broadleaf-and-mixed forest, as predicted. The recent detection of Asian gypsy moth in Texas suggests this insect may also survive in areas with temperate grasslands and savannas.

Survey

European and Asian strains of gypsy moth respond to the same pheromone (Cardé et al. 1996): cis-7R,8S-epoxy-2-methyloctadecane, commercially known as disparlure. Standard protocols for the survey of European gypsy moth using pheromone-based traps also apply to Asian gypsy moth. Detailed protocols are provided by USDA (2006).

Asian forms of gypsy moth are attracted to lights, especially UV lights (Wallner et al 1995). As a result, a monitoring program could be developed using light traps, but the cost of operating the trap and sorting the volume of insects that are captured make this approach less desirable.

Asian and European strains of gypsy moth differ in mitochondrial DNA (Bogdanowicz et al 1993) and nuclear DNA. Genetic differences can be most easily resolved based on polymerase chain reaction (PCR)-based analysis of randomly amplified polymorphic DNA (Garner and Slavicek 1996, Schreiber et al. 1997), amplified fragment length polymorphisms (Reineke et al. 1999), microsatellite DNA (Bogdanowicz et al. 1997), or ribosomal DNA (Pfeifer et al. 1995). In a comparison of RAPDs and restriction site polymorphisms of ribosomal DNA, Reineke and Zebitz (1999) concluded that both methods worked well to distinguish Asian from European populations of gypsy moth. The analysis of ribosomal DNA may be slightly better able to resolve subtle differences in geographic origin (i.e., European *vs.* North American). The results from RAPD analysis can vary from lab to lab due to subtle deviations from standard protocols, but Reineke and Zebitz (1999) were able to reproduce the results of earlier authors.

Certain morphological features may be used to separate strains. Detailed image analysis of head capsule color has been used to separate subpopulations of European and Asian gypsy moth larvae (Wallner et al. 1994). Asian and European strains may also be identified based on distinct patterns in wing venation (Kunkel 1995). The utility of these approaches depends more heavily on the stage and condition of a specimen than do the molecular methods.

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Lymantria mathura[*]

Scientific Name

Lymantria mathura Moore

Fig. 1. Adult female *Lymantria mathura*.
[Image from David Mohn, www.forestryimages.org]

Synonyms:
Portheria mathura (Moore)
Ocneria mathura (Moore)
Lymantria aurora Butler
Lymantria fusca Leech
Lymantria mathura aurora Butler

Common Names

Pink gypsy moth
Rosy gypsy moth

Type of Pest

Moth, defoliator

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera, **Family:** Lymantriidae

Reason for inclusion in manual

CAPS Priority Pest (FY 2007)

Pest Description

"*Lymantria mathura* Moore (Lepidoptera: Lymantriidae) is a moderate sized moth... There is marked sexual dimorphism in size and colour. The male is smaller (wing expanse male: 35-50 mm [~1½-2 in]; female: 75-95 mm [~3-3¾ in]), with the forewings brown and hindwings yellow. In females the forewings are white with dark markings, and the hindwings pink..." (Fig. 1) (Roonwal 1979a).

"[Male] Upperside-fore wing greyish white, markings brown, with pale-brown interspaces; with two or three black and yellow spots at the base; two transverse subbasal irregular lines, between which is a broad band; a round spot within the cell and a blackish curved streak at its end; three transverse discal lunulated bands, the first broad, the others narrow; a marginal row of spots: hind wing dull yellow, with a blackish discal spot, narrow submarginal maculated band, and a marginal row of small spots. Underside dull yellow, suffused with pale brown

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[*] This document is largely excerpted from the report: Davis, E.E., S. French, and R.C. Venette. 2005. Mini-Risk Assessment: Pink gypsy moth, *Lymantria mathura* Moore [Lepidoptera: Lymantriidae]. Available on-line at www.aphis.usda.gov/ppq/ep/pestdetection/pra/lmathurapra.pdf

between the veins, with darker-brown discal and marginal spots. Thorax white, with yellow and black spots. Abdomen yellow, tuft white, with dorsal, lateral, and a row beneath of black spots. Head at the sides, palpi in front, and legs yellow; palpi above and at the sides, and spots on the legs, black. Antennae brown. Expanse 2¼ inches" (Moore 1865).

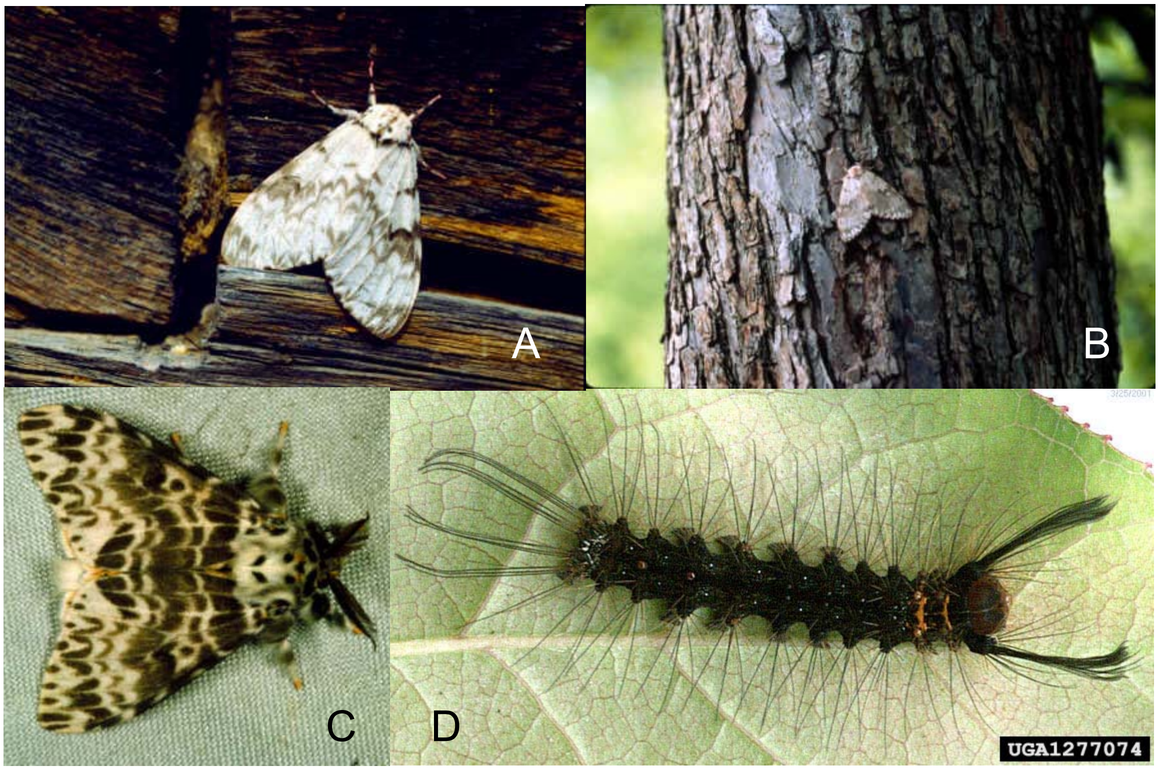

Fig. 2. *Lymantria mathura*: (A&B) Adult female with pink hind wings; (C) adult male with yellow hind wings ; (D) larva on foliage of deciduous host. Images not to scale.
[Images (A-C) from W. Wallner, http://www.inspection.gc.ca/english/sci/surv/data/lymmate.shtml
(D) David Mohn, www.forestryimages.org]

"[Egg-masses and covering hairs] Egg masses are laid from ground-level up to about 18 m [60 ft.] of the trunk, but are most dense between the levels of 0.5 to 5 m [1½-16 ft]. They are flat, of an ovoid-elongate or other shape, with irregular edges, and vary in extent from about 0.5 x 1 cm to 6 x 15 cm. From a distance the egg masses are visible as characteristic white, fluffy patches against the dark-coloured bark. Each egg-mass contains about 50 to 1,200 or more eggs which are laid 2 to 4 layers deep directly on the bark. An egg-mass is covered over with a nearly one-millimetre, white thick felt-like covering composed of long, white, silken hairs (... these hairs are shed by the female from the anal tuft. ...). The hairs are about 800-1200 µ long and 3.1-6.2 µ in diameter; one end is knob-like, the other pointed; a few such hairs are also mixed with the eggs. Freshly laid eggs are rounded, have a flat base, the maximum and minimum diameters varying from 1.13-1.19 mm and 0.86-0.92 mm respectively" (Roonwal 1979a).

"[Egg-mass after hatching] After the majority of eggs have hatched, an egg mass presents a changed appearance. Firstly, the hair-covering which has hitherto (for several months in the case of the overwintering eggs) remained pure white, now becomes dull-coloured, a dirty cream, and, in a few cases, with irregular patches of pale buff. Secondly, the hair covering is pierced by numerous rounded holes of varying diameters (c. 0.5-3 mm) through which the newly hatched larvae have escaped. Beneath the thin, hole-pierced, hairy covering, there is a flat, hollow space containing the remnants of eggshells and a few remaining eggs which have not yet hatched" (Roonwal 1979a).

"[Larvae] Three main colour forms are found in mature caterpillars, the following proportions being noticed in 1,613 caterpillars examined: grey-white 66%, intermediate 11%, and blackish brown 23%. The details of colour are described below briefly.

> Form I (Grey-white): Ground colour dirty white tinged with grey. Dorsal: Head white with numerous black or brown spots; frons with a longitudinal median black streak; rest of body grey-white, with numerous fine dots forming paired patches. A transverse yellow-brown streak present between pro- and mesothorax, and another in middle of metathorax: abdominal warts blackish; paired lateral papules on abdomen white, with tufts of long white and brown hairs. Long pencil-like plumes of hairs on head and on, end of abdomen black. Ventral: Brownish pink; legs and prolegs brown, the latter with a black patch externally.
>
> Form II (Intermediate): Dorsal: Ground colour pale brown, with a median white patch on abdominal terga 4 and 5. Ventral: As in Form I.
>
> Form III (Blackish brown): Dorsal: Ground-colour dark brown to almost black; numerous black spots visible in brown larvae but merged with ground-colour in darker ones; several small white dots present on abdominal terga 4 to the last, and large white patches on terga 4-6. Ventral: Ashy, suffused with a little pink in the median parts; rest as in Form I.
>
> In the masses of caterpillars on tree trunks the various colour types are mixed on individual trees; this fact has a protective value by making detection by enemies difficult" (Roonwal 1979a).

"The size ... characteristics of the six larval stages are given below briefly...

> Stage I. Length 3 mm; head-width 0.5 mm. Generally black dorsally; meso- and metathorax and segment 5 of abdomen brown; legs black; prolegs pale brown with a black patch externally.
>
> Stage II. Length 5 mm; head-width 0.7 mm. Generally black dorsally; meso- and metathorax greyish; last abdominal segment pale brown with blackish tinge; rest as in Stage I.

> Stage III. Length 13 mm; head-width 1.5 mm. Head brown; body black above, paler below; thoracic terga with yellow-brown spots; legs black, prolegs brown with a black external patch.
>
> Stage IV. Length 20 mm; head-width 2.5 mm. Head above either black (brown distally) or pale green with black dots; sides brown; body black with white warts; meso- and metathorax with brown stripes anteriorly; legs and prolegs as in Stage III.
>
> Stage V. Length 30-40 mm [~1-1/2 in]; head-width 3.5 mm. Head above brown to grey, speckled with black; body black with many minute white spots; pro- and mesothorax with a transverse brown streak at the distal edge; ninth abdominal segment with a pair of prominent dorsal white spots; legs and prolegs reddish brown, the latter with a large black patch externally.
>
> Stage VI. Length 60-85 mm [~2 1/3-3 1/3 in]; head-width 5-6 mm. With sexual dimorphism, females being longer (males: 60-65 mm, females: 70-85 mm). Colour pattern similar to Stage V, but in ground-pattern three types recognizable, viz., grey-white, blackish-brown and intermediate (vide infra). Older larvae well 'camouflaged' against tree trunks" (Roonwal 1979a).

"[Pupa] The pupa is of the 'obtect adecticus type,' and the appendages are firmly soldered to the body. It is buff to dark brown, about 20-36 mm [¾-1 ½ in] long, and shows sexual dimorphism; the female pupa is paler, larger and heavier than the male, as follows:

> Female: Buff to pale brown. Length (including hair tufts) 30-36 mm; maximum width 10-14 mm. Weight 0.88 gm (average of 18 pupae).
>
> Male: Very dark chocolate brown, Length (including hair tufts) 15-25 mm; maximum width 6-8 mm. Weight 0.14 gm (average of 53 pupae)" (Fig. 2) (Roonwal 1979a).

Biology and Ecology

In its native range, *L. mathura* is univoltine or bivoltine (Beeson 1941, Browne 1968, Roonwal 1979a, Baranchikov et al. 1995, Lee and Lee 1996). The first generation occurs between April and October. Flight activity is not well known for this species, but is thought to coincide with peak flight activity of two closely related species, *L. dispar* and *L. monacha* (Anon. 2001). Males are scarcely seen and die about a week before females. Females congregate in groups of 6 or more near egg masses and become inactive after laying eggs (Roonwal 1979a).

Eggs are laid between mid-April and mid-June and hatch in 3-4 weeks. Between 50-1,200 eggs are laid in white, distinctive silky hair-covered masses on trunks and large branches of deciduous hosts (Browne 1968, Roonwal 1979a). Eggs are laid from the base of a tree trunk to a height of about 18 m [60 ft], and most egg masses tend to occur at a height between 0.5-5 m [1½-16 ft] (Roonwal 1979a).

Larvae occur from early June to late September. After eggs hatch, the egg mass becomes darker in color. The group of newly hatched larvae remains near the hair-covered mass for 2-3 weeks. It is not known whether the larvae receive some nutritive benefit from the mass prior to feeding on foliage (Roonwal 1979a). The insect progresses through six instars. Early instar *L. mathura* larvae possess the ability to disperse by dropping on a trailing silk thread and utilizing air and wind currents to "balloon" to other locations (Zlotina et al. 1999). *Lymantria mathura* larvae may disperse farther than *L. dispar* via wind (Zlotina et al. 1999). Late instar larvae exhibit a diurnal feeding behavior. Although they are inactive during most daylight hours, late instars begin to twist distinctively before dusk, then crawl to the tree crown to feed until dawn, followed by a rapid descent to the trunk (Roonwal 1979a, Zlotina et al. 1999). Density on the host trunk reached a maximum at 5 PM, just prior to the evening migration to the crown Roonwal (1979a) Larval densities can average 1,338/tree (range 1,140-1,671) (Roonwal 1979a).

Pupae are present from late July to late October. Pupation often occurs in groups of 40-50 in protected areas of branches, in leaf litter at the base of trees, or on the back or underside of signs or other objects (Browne 1968, Roonwal 1979a).

In the second or overwintering generation, eggs are laid between early September to mid-October, and embryos develop within 6 weeks. This generation overwinters as developed embryos within eggs which hatch between February and early April, depending on temperature.

In outbreak years, *L. mathura* tends to lay eggs on many tree species, including non-hosts. *Lymantria mathura* eggs were laid on 185 different host species, and of these, 22 tree species were later defoliated by feeding larvae, and 6 species were heavily defoliated [see 'Known Hosts']. *Lymantria mathura* has historically demonstrated food preferences, but these preferences depend on which hosts are available (Roonwal 1979a, Baranchikov et al. 1995). The selection of a location for egg deposition may also depend on the presence or density of other egg masses, host preference, and the extent of feeding that has already occurred on a host (Roonwal 1979a).

When population density is high, parasitism by hymenopterans or infection by polyhedral viral disease may result in high mortality of larvae and pupae (Roonwal 1979a).

Pest Importance

Lymantria mathura is a major defoliator of deciduous trees in the Palearctic, primarily in eastern Asia from India to the Russian Far East (Roonwal 1979a, Baranchikov et al. 1995, reviewed in CAB 2004, reviewed in EPPO 2005). Spurred by concerns surrounding *L. mathura*, the US Department of Agriculture-Animal and Plant Health Inspection Service, USDA Forest Service and Russian counterparts have developed an early warning system to alert US pest officials about periods of increased insect activity and prevent the introduction of this insect (Anon. 2001). US officials are also alerted when New Zealand finds a Russian freighter to be infested with this insect (reviewed in USDA 2001).

In India, *L. mathura* is an economically important forest pest, which defoliates *Shorea robusta*, and several other deciduous forest and fruit tree species [see 'Known Hosts']. Roonwal (1953, 1962, 1979a) states that outbreaks are periodic, and prior to the worst epidemic of this pest on record in India during 1953, *L. mathura* was considered unimportant. The outbreak extended from the western sub-Himalayas to West Bengal, encompassing several adjacent forest divisions. In the Russian Far East, there has been only one reported outbreak in the Primorie region, where losses amounted to hundreds of hectares of deciduous forests (Baranchikov et al. 1995). Damage to chestnut resulted from an outbreak of *L. mathura* in areas of Kyonggi province, Korea (Lee and Lee 1996).

Establishment of *L. mathura* in the US could also adversely impact trade. This insect has been proposed as an A2 quarantine pest in Europe, a status reflecting its limited presence (EPPO 2005). Potentially infested products within the US could become the focus of domestic or international quarantines.

Synthetic insecticides are an option for control, but in many natural settings, complex terrain limits the feasibility of this option, especially over large areas. However, as has been observed with *L. dispar*, formulations of endotoxin from *Bacillus thuringiensis* (e.g., *Bt-k*) may be applied aerially to localized populations (Myers and Hosking 2002). *Bt* is generally considered host specific (Lacey and Siegel 2000), but some exceptions have been noted especially after repeated applications (Lacey and Siegel 2000, Boulton 2004). Biological control is a much more likely option (Rosovsky 2001). Previous experience with gypsy moth demonstrates that predators, parasitoids, and pathogens might be introduced. In previous years, generalist agents (e.g., *Compsilura concinata*) were introduced, often with significant impacts on non-target species (reviewed in Syrett 2002). Current protocols for the screening of agents limit the likelihood of these severe impacts to non-target species (reviewed in Hoddle and Syrett 2002).

Risks associated with *L. mathura* have been evaluated previously. In the Exotic Forest Pest Information System, *L. mathura* was considered to pose a very high risk to North America forests relative to other forest pests and pathogens, and this assessment was given with a very high degree of certainty (Rosovsky 2001).

Gninenko and Gninenko (2002) proposed a scoring system to evaluate the relative propensity of different lymantriids to be moved by international shipping. These authors suggest that *L. mathura* is less likely than *L. dispar* or *L. monacha* to be moved by shipping, but it is more likely to be moved than 26 other species of Lymantriidae. Limited biological information about lymantriids of the Russian Far East, including *L. mathura*, complicates the assessment of risk (Gninenko and Gninenko 2002).

Symptoms

Lymantria mathura larvae are gregarious defoliators, able to consume whole leaves and sometimes avoid tough veins in older foliage growth. Larvae may also feed on flowers and tender young shoots (Browne 1968, Roonwal 1979a). Damage of this nature can result in decline in overall growth and development, a reduction in yield or total crop loss (fruit crops), or even tree death (Singh 1954, Roonwal 1979a).

Egg masses or larvae may also be evident on trees (reviewed in Rosovsky 2001).

Known Hosts

Lymantria mathura is a polyphagous pest of taxonomically diverse deciduous trees (Appendix B). *Lymantria mathura* reportedly feeds on more than 45 genera in 24 families. Numerous accounts of preferential feeding are reported, but these reports are somewhat inconsistent (Roonwal 1979a, Baranchikov et al. 1995).

Hosts	**References**
Aphanamixis polystachya (=*Amoora* or "*Ammora*" *rohituka*) (rayana)[1]	(Roonwal 1979a)
"Catania" sp.[1]	(Lee and Lee 1996)
Abies nephrolepis (=*A.* "*nephroletis*") (fir, Manchurian)[1, 2]	(Zlotina et al. 1998)
Abies sp. (fir)	(Rosovsky 2001, CAB 2004)
Acrocarpus fraxinifolius (pink-cedar)	(Roonwal et al. 1962, Roonwal 1979a)
Alnus sp. (alder)	(Wallner et al. 1995, Yamazaki and Sugiura 2004)
Alstonia scholaris (blackboard tree)	(Roonwal 1979a)
Anogeissus lalifolia (dhaoda)	(Roonwal 1979a)
Artocarpus lacucha (=*A. lakoocha*) (monkey-jack tree)	(Roonwal 1979a)
Betula sp. (birch)	(Baranchikov et al. 1995, Wallner et al. 1995, Zlotina et al. 1998, Rosovsky 2001, CAB 2004)
Butea monosperma (Bengal kino)	(Roonwal 1979a)
Carya sp. (hickory)	(Rosovsky 2001, CAB 2004)

Hosts	**References**
Castanea mollissima (chestnut, Chinese hairy)	(Rosovsky 2001, CAB 2004)
Castanea sativa (chestnut, European)	(Roonwal 1979a)
Castanea sp. (chestnut)	(Zhang 1994, Lee and Lee 1996, Rosovsky 2001, CAB 2004)
Dimocarpus longan (longaan)	(Mohn 1993)
Duabanga grandiflora (=*D. sonneratioides*) (duabanga)	(Roonwal 1979a)
Elaeodendron (=“*Eeodendron*” *glaucum*) (Ceylon tea)[1]	(Roonwal 1979a)
Fagus sp. (beech)	(Mohn 1993, Pucat and Watler 1997, Zlotina et al. 1998, Gries et al. 1999, Rosovsky 2001, CAB 2004)
Fagus grandifolia (beech, American)[2]	(Zlotina et al. 1998)
Fagus sylvatica (beech, European)[2]	(Zlotina et al. 1998)
Ficus benghalensis (Indian banyan)	(Roonwal 1979a)
Fraxinus sp. (ash)	(Rosovsky 2001, CAB 2004)
Grewia sapinda	(Roonwal 1979a)
Haldina cordifolia (=*Adina cordifolia*) (haldu)	(Roonwal 1979a)
Hippophae rhamnoides (sea buckthorn)[2]	(Baranchikov et al. 1995)
Juglans mandshurica (walnut, Manchurian)	(Baranchikov et al. 1995, Zlotina et al. 1998)
Juglans sp. (walnut)	(Rosovsky 2001, CAB 2004)
Larix sp. (larch)	(Wallner et al. 1995, Rosovsky 2001, CAB 2004)
Liquidambar formosana (Formosan sweetgum)	(Mohn 1993, Zhang 1994, Rosovsky 2001, CAB 2004)
Litchi chinensis (lychee)	(Singh 1954, Roonwal 1979a, Rosovsky 2001, CAB 2004)
Mallotus philipinensis (kamala)	(Roonwal 1979a)
Malus sp. (apple)	(Mohn 1993, Pucat and Watler 1997, Zlotina et al. 1998, Gries et al. 1999, CAB 2004, Yamazaki and Sugiura 2004)
Malus mandshurica (=*M.* “*mandjurica*”) (crabapple, Manchurian)[1]	(Baranchikov et al. 1995)
Malus prunifolia (=*M.* “*pruniflora*”) (apple, Chinese)[1, 2]	(Baranchikov et al. 1995)
Manchurian nut	(Yurchenko and Turova 2002)

Hosts	**References**
Mangifera indica (mango)	(Singh 1954, Browne 1968, Roonwal 1979a, Mohn 1993, Pucat and Watler 1997, Zlotina et al. 1998, Rosovsky 2001, CAB 2004)
Melia azedarach (china berry tree)	(Roonwal 1979a)
Millettia pinnata (=*Pongamia glabra*) (pongame oil tree)	(Roonwal 1979a)
Morus alba (mulberry, white)	(Roonwal 1979a)
Neolamarckia cadamba (=*Anthocephalus cadamba*) (kadam)	(Browne 1968, Roonwal 1979a, Pucat and Watler 1997, Rosovsky 2001, CAB 2004)
Pinus koraiensis (pine, Korean)[2]	(Zlotina et al. 1998)
Pinus sp. (pine)	(Lee and Lee 1996, Rosovsky 2001, CAB 2004)
Populus sp. (cottonwood)	(Baranchikov et al. 1995, Zlotina et al. 1998)
Prunus cerasoides (=*P. puddum*) (cherry, wild Himalayan)	(Roonwal 1979a)
Prunus sp. (stone fruit)	(Mohn 1993, Pucat and Watler 1997, Zlotina et al. 1998, CAB 2004, Yamazaki and Sugiura 2004)
Pseudotsuga menziesii (fir, Douglas)	(Rosovsky 2001, CAB 2004)
Pterygota alata (=*Sterculia alata*) (Buddha's coconut)	(Roonwal 1979a)
Pyrus sp. (pear)	(Pucat and Watler 1997, Zlotina et al. 1998, CAB 2004)
Quercus sp. (oak)	(Odell et al. 1992, Mohn 1993, Wallner et al. 1995, Lee and Lee 1996, Pucat and Watler 1997, Zlotina et al. 1998, Gries et al. 1999, Rosovsky 2001, CAB 2004, Yamazaki and Sugiura 2004)
Quercus acuta (oak, Japanese evergreen)	(Wileman 1918)
Quercus alba (oak, white)[2]	(Zlotina et al. 1998)
Quercus dentata (oak, Daimyo)	(Wileman 1918)
Quercus glauca (oak, ring-cup)	(Funakoshi 2004)
Quercus leucotrichophora (=*Q. incana*) (oak, banj)[3]	(Beeson 1941, Roonwal 1953, Roonwal et al. 1962, Browne 1968, Roonwal 1979a, Pucat and Watler 1997, Rosovsky 2001, CAB 2004)

Hosts	**References**
Quercus mongolica (oak, Mongolian)	(Baranchikov et al. 1995, Zlotina et al. 1998, Rosovsky 2001, Yurchenko and Turova 2002, CAB 2004)
Quercus prinus (oak, chestnut)[2]	(Zlotina et al. 1998, Gries et al. 1999)
Quercus serrata (=*Q. glandulifera*) (oak, Konara)[3]	(Wileman 1918, Beeson 1941, Roonwal 1953, Roonwal et al. 1962, Browne 1968, Roonwal 1979a, Pucat and Watler 1997, Rosovsky 2001, CAB 2004)
Quercus variabilis (oak, Chinese cork)[2]	(Zlotina et al. 1998)
Rhus sp. (sumac)	(Gries et al. 1999)
Rosa rugosa (rose, Japanese)[2]	(Baranchikov et al. 1995)
Salix sp. (willow)	(Zlotina et al. 1998, Rosovsky 2001, CAB 2004)
Salix fragilis (willow, crack)[2]	(Baranchikov et al. 1995)
Shorea robusta (sal tree)[3]	(Beeson 1941, Roonwal 1953, Roonwal et al. 1962, Browne 1968, Roonwal 1979b, 1979a, Pucat and Watler 1997, Rosovsky 2001, CAB 2004)
Syzigium cumini (=*Eugenia jambolana*) (plum, Java)[3]	(Beeson 1941, Roonwal 1953, Roonwal et al. 1962, Browne 1968, Roonwal 1979a, Pucat and Watler 1997, Rosovsky 2001, CAB 2004)
Terminalia arjuna (arjuna)[3]	(Beeson 1941, Roonwal 1953, Roonwal et al. 1962, Browne 1968, Roonwal 1979a, Pucat and Watler 1997, Rosovsky 2001, CAB 2004)
Terminalia belerica (beleric)	(Roonwal 1979a)
Terminalia elliptica (=*T. tomentosa*) (asna)	(Roonwal 1979a)
Terminalia myriocarpa (hollock)[3]	(Beeson 1941, Roonwal 1953, Roonwal et al. 1962, Browne 1968, Roonwal 1979a, Pucat and Watler 1997, Rosovsky 2001, CAB 2004)
Terminalia pyrifolia	(Roonwal 1979a)
Tilia mandshurica (linden, Manchurian)	(Zlotina et al. 1998)
Toona ciliata (=*Cedrela toona*) (Australian red-cedar)	(Roonwal 1979a)
Toxicodendron succedaneum (=*Rhus succedanea*) (waxtree, Japanese)	(Wileman 1918)

Hosts	References
Ulmus sp. (elm)	(Baranchikov et al. 1995, Zlotina et al. 1998)
Ulmus davidiana (elm, Japanese)	(Yurchenko and Turova 2002)
Zelkowa sp. (zelkowa)	(Gries et al. 1999)
Zelkowa acuminata (zelkowa, Japanese)	(Wileman 1918)

1. Likely misspelling in literature, or unrecognized name.
2. Experimental hosts (Baranchikov et al. 1995, Zlotina et al. 1998)
3. A preferred host species, noted as heavily defoliated among 185 tree species with egg masses and 22 tree species with some defoliation (Roonwal 1979a).

Known Distribution

Lymantria mathura is present throughout much of Asia.

Locations	References
Bangladesh	(Rosovsky 2001, CAB 2004)
China	(Wileman 1918, Lewis et al. 1984, Odell et al. 1992, Mohn 1993, Zhang 1994, Wallner et al. 1995, Pucat and Watler 1997, Zlotina et al. 1998, Gries et al. 1999, Rosovsky 2001, CAB 2004, Khrimian et al. 2004, Schintlmeister 2004)
India	(Wileman 1918, Beeson 1941, Sevastopulo 1947, Roonwal 1953, Singh 1954, Roonwal et al. 1962, Browne 1968, Roonwal 1979a, 1979b, Zhang 1994, Baranchikov et al. 1995, Pucat and Watler 1997, Zlotina et al. 1998, Gries et al. 1999, Rosovsky 2001, CAB 2004, Khrimian et al. 2004, Schintlmeister 2004)
Japan	(Wileman 1918, Zhang 1994, Pucat and Watler 1997, Zlotina et al. 1998, Gries et al. 1999, Rosovsky 2001, Funakoshi 2004, Khrimian et al. 2004, Schintlmeister 2004, Yamazaki and Sugiura 2004)
Kashmir	(Wileman 1918)
Korea	(Wileman 1918, Lee and Lee 1996, Rosovsky 2001)
Korea, Republic of	(CAB 2004)
Kurile Islands	(Wileman 1918)
Myanmar (formerly Burma)	(Roonwal 1979a)

Locations	**References**
Pakistan	(Browne 1968, Pucat and Watler 1997, Rosovsky 2001)
Russia	(Wileman 1918, Baranchikov et al. 1995, Pfeifer et al. 1995, Wallner et al. 1995, Zlotina et al. 1998, Zolotarenko and Dubatolov 1998, Gries et al. 1999, Oliver et al. 1999, Zlotina et al. 1999, Anon. 2001, Rosovsky 2001, CAB 2004, Khrimian et al. 2004)
Taiwan	(Zhang 1994, Pucat and Watler 1997, Gries et al. 1999, Rosovsky 2001, CAB 2004, Schintlmeister 2004)
temperate broadleaf and mixed forest[1]	(Schintlmeister 2004)
temperate coniferous forest[1]	(Schintlmeister 2004)
tropical and subtropical dry broadleaf forest[1]	(Schintlmeister 2004)
tropical and subtropical moist broadleaf forest[1]	(Schintlmeister 2004)
United States of America (N. America; west coast ports)[2]	(Baranchikov et al. 1995, CAB 2004)

1. Refer to map by Schintlmeister for general locations; no scale provided (Schintlmeister 2004)
2. Intercepted but not established (Baranchikov et al. 1995, CAB 2004)

Potential Distribution within the US

In general, *L. mathura* occurs in cool, temperate to warm climates with variable seasonal rainfall and dry periods. The currently reported distribution of *L. mathura* suggests that the pest may be most closely associated with biomes characterized as: temperate broadleaf and mixed forests; temperate coniferous forests; tropical and subtropical dry broadleaf forests; and tropical and subtropical moist broadleaf forests. Of these biomes, only tropical and subtropical dry broadleaf forests do not occur in the US. Consequently, approximately 38% of the continental US would have a suitable climate for *L. mathura*.

Survey

Several tools are available to assist with surveys for *L. mathura*. Pheromone-baited traps are particularly useful for regional surveys while visual inspections are necessary for conveyances that may be bringing *L. mathura* into an area. Inspectors should look for egg masses on any products originating from infested areas. Egg masses may be deposited on logs, nursery stock, forest products, or sea containers (Pucat and Watler 1997). Females prefer to deposit eggs on a rough surface (Roonwal 1979a).

Sex pheromones for *L. mathura* have been identified and can be used for detection surveys. Early research (reviewed in Gries et al. 1999) indicated that males of *L. mathura* were attracted to *cis*-7,8-epoxy-2-methyloctadecane and 2-methyl-*Z*7-octadecene (Odell et al. 1992). Males also demonstrated electrophysiological responses to (*Z*3,*Z*6,*Z*9)-nonadecatriene and (9*S*,10*R*)-9,10-epoxy-*Z*3,*Z*6-nonadecadiene in extracts from abdominal tips of *L. mathura* females (Oliver et al. 1999). Subsequent research revealed that major sex pheromone components include a blend of (9*R*,10*S*)-*cis*-9,10-epoxy-*Z*3,*Z*6-nonadecadiene (named (+)-mathuralure) and (9*S*,10*R*)-*cis*-9,10-epoxy-*Z*3,*Z*6-nonadecadiene (named (-)-mathuralure) in a 1:4 ratio (Gries et al. 1999). Neither component is attractive alone (Gries et al. 1999). Khrimian et al. (2004) explain that the enantiomer (-)-mathuralure is equivalent to the compound identified by Oliver et al. (1999) and provide a detailed protocol for the synthesis of (+)-mathuralure and (-)-mathuralure in a 4:1 ratio. The pheromone is most effectively deployed using PVC-coated string dispensers with 64 μg pheromone per cm (Khrimian et al. 2004). Traps baited with (+)-disparlure will also attract male *L. mathura* (Odell et al. 1992).

Pheromone lures have been used with Delta sticky traps (Fig. 3, Gries et al. 1999) or 3.8-L milk carton traps (Odell et al. 1992). Traps are generally hung 1.5-2 m [ca. 5-6.5 ft] above ground (Odell et al. 1992, Gries et al. 1999). To improve diffusion of the pheromone, traps have been suspended 0.6 m [2 ft] from the trunk of a tree on wooden stakes nailed to the tree (Odell et al. 1992). For research purposes, traps were placed 20-25 m apart (Gries et al. 1999), but standard protocols for detection of gypsy moth in uninfested states should be appropriate.

Wallner et al. (1995) evaluated several light sources (e.g., diffuse coated sodium lamps; phosphor-coated, high-pressure mercury lamps, and blacklight lamps) and found that *L. mathura* were most attracted to blacklight. However, light traps are generally considered ineffective and impractical for regional monitoring of this insect (reviewed in CAB 2004).

Lymantria mathura is not likely to be confused with other lymantrids, particularly if a specimen is an adult or late instar larva (reviewed in EPPO 2005). Eggs or neonates are incredibly difficult to distinguish, and molecular tools are being developed to aid with identification (Armstrong et al. 2003).

Fig. 3. Delta trap used for detecting lymantriids.
[Image from USDA APHIS PPQ Archives, www.forestpests.org]

Lymantria mathura might be confused with *L. monacha* (also exotic, not known to occur in the US) or *L. dispar*.

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Spodoptera littoralis[*]

Scientific Name

Spodoptera littoralis Boisduval

Synonyms:
At the generic level:
Spodoptera Guenée 1852; (Viette 1863)

- *Prodenia* Guenée 1852a; (Hampson 1894)
- *Hadena* Boisduval (1833)

At the species level:
littoralis Boisduval 1833

- *testaceoides* Guenée 1852
- *retina* Guenée 1852

Common Names

Egyptian cotton leafworm

Type of Pest

Moth, defoliator

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for inclusion in manual

CAPS Priority Pest (FY 2007)

Pest Description

Spodoptera littoralis and *S. litura*, another exotic lepidopteran pest, have long been “taxonomically confused” (Mochida 1973). Because of their morphological similarities, the two species were erroneously considered as a single species in historical literature (Hafez and Hassan 1969, reviewed in CABI/EPPO 1997). Both species are difficult to distinguish without close examination of the genitalia (Mochida 1973, Brown and Dewhurst 1975, reviewed in DEFRA 1999). With regard to geographical distribution, “the ranges of the two species do not currently overlap and neither has extended its range (except in the special case of glasshouses in Europe)” (reviewed in CABI/EPPO 1997). Unlike *S. littoralis*, *S. litura* is found in much of southeastern Asia and Oceania (IIE 1993).

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[*] This document is largely excerpted from the report: Venette, R.C., E.E. Davis, J. Zaspel, H. Heisler, and M. Larson. 2003. Mini-Risk Assessment: Egyptian Cotton Leafworm, *Spodoptera littoralis* Boisduval [Lepidoptera: Noctuidae]. Available on-line at www.aphis.usda.gov/ppq/ep/pestdetection/pra/tleucotretapra.pdf

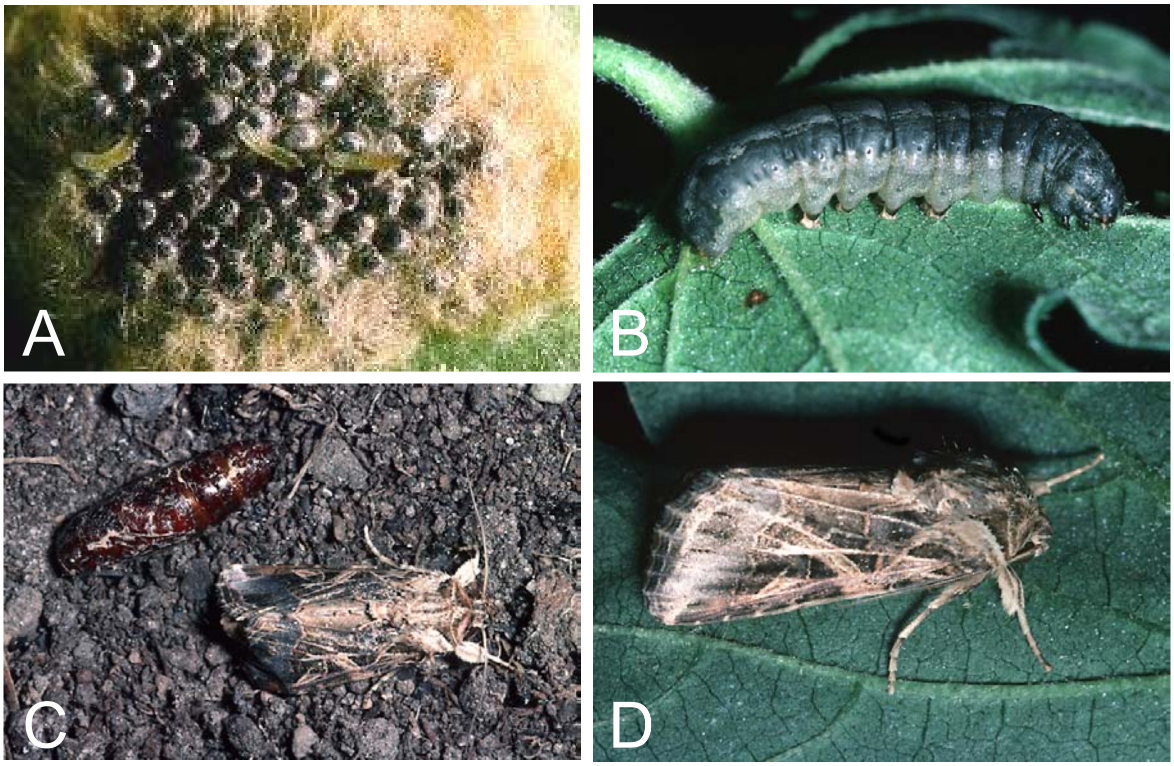

Fig. 1. Life stages of *Spodoptera littoralis*, images not to scale: (A) neonates and egg mass covered in scales from female; (B) late instar larva; (C) pupa and adult on soil; (D), adult on leaf
[Image A from http://www.defra.gov.uk/planth/pestnote/spod.htm; B-D from Entopix]

Spodoptera species can be recognized by the gray to brown forewing ground color and white hindwing (Pogue 2002). Several forewing patterns exist and once recognized can distinguish *Spodoptera* from other genera. External color and pattern are almost indistinguishable from *S. litura* (Pogue 2002). Forewings (length=12-16 mm) are brownish with a pale yellow along the median vein and the hindwing vein tips of *S. littoralis* are white. The ochreous median area of the forewing is between the antemedial and postmedial lines, and below vein M (Fig. 2).

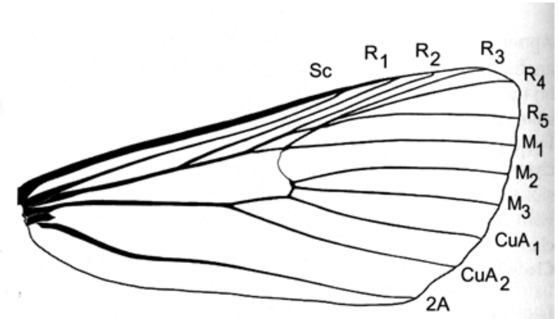

Fig. 2. General diagram of forewing venation
[Reproduced from Pogue (2002)]

Forewing: male: Length 12-16mm; ground color brown; basal line absent; longitudinal black dash at base absent (Pogue 2002).

Forewing female: Length, 13-16mm. Longitudinal black dash at base absent (Pogue 2002).

Variation: Variation can be found in the forewing ground color of this species and male external morphology is somewhat different from the female. Also, the orbicular spot is larger in the male than in the female (Pogue 2002).

Hindwing: Ground color white; no spot on underside.

Larva: The head and frons are brown and the cutting edge of the mandible is serrate (Pogue 2002). The pronotum dorsolateral stripe is absent or inconspicuous, with a few faded spots at the margins and the mid-dorsal stripe is narrower than the dorsolateral stripe. The mesothoracic segmental spot is round with a white spot at the base of the segmental spot with the lateral dark spot absent. The abdomen is smooth, with an inconspicuous middorsal stripe, narrower than dorsolateral stripe. Segmental spots on abdominal segments 7 and 8 larger than on 1-6; wide on segments 1 and 8, almost extending to middorsal stripe with a white spot mid-basally in segmental spot. Segment 1 without lateral dark spot and segments 2-6 without lateral dark spots in spiracular band. Dorsolateral stripe yellow, and spiracles with black border and brown center. Subspiracular stripe continuous through abdominal segment 1 (Pogue 2002).

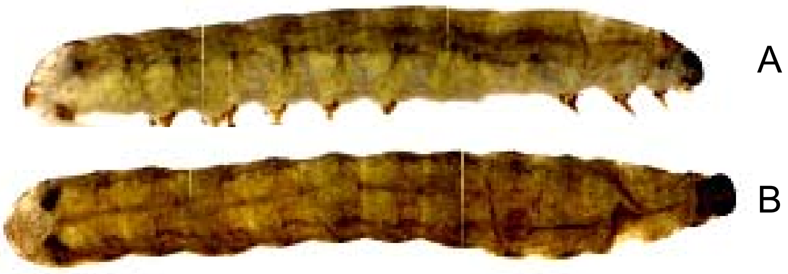

Fig. 3. Larva of *Spodoptera littoralis*; A: Lateral view, B: Dorsal view.
[Reproduced from Pogue (2002)].

Other detailed, descriptive features are provided by Pogue (2002).

Biology and Ecology

This insect is multivoltine with three to seven generations annually depending on climatic conditions (reviewed in USDA 1982). The complete life cycle (from egg to egg) requires about 25 days in temperate summers, but generation time is longer as temperatures cool. A total of 392 degree days [°C] above a threshold of 10.5°C [51°F] is needed to complete a generation (reviewed in USDA 1982). Females are not long-lived, lasting only 2-22 days depending on season. In the summer, females live less than a week, but in a Mediterranean winter may live up to three weeks (reviewed in USDA 1982). Males do not live as long as females. Adults are particularly active at twilight and during the night. Moths are more active when relative humidity is high. Females mate shortly after emergence (reviewed in USDA 1982). A female will produce 40-3600 (average 3158) eggs, depositing most of these eggs within 4 days after mating (Duodu and Sham 1986-87). Adults may disperse up to 1.5 km [~1 mi] in a 4 hr period (reviewed in CAB 2000).

Eggs will begin to hatch in about 3 days (Duodu and Sham 1986-87). An egg is white when it is first deposited but will turn black if it is non-viable; viable eggs become opaque within a day (Duodu and Sham 1986-87).

As the insect develops, it completes six instars. Early instars remain on the underside of leaves and feed throughout the day (reviewed in CAB 2000). Third or fourth instars remain on a plant but do not feed during daylight; later instars migrate off the plant to rest in the soil during the day and return at night (reviewed in CAB 2000).

The insect pupates in the soil or leaf litter (Duodu and Sham 1986-87). Pupation lasts 5-31 days depending on temperature (reviewed in USDA 1982).

For a more detailed description of the biology of *S. littoralis*, see Ellis (2003).

Pest Importance

Spodoptera littoralis is a polyphagous pest that poses a high degree of risk to US agriculture and ecosystems. In a pathway-initiated risk assessment, this insect was judged highly likely of establishing in the US should it be introduced; the consequences of its establishment were considered severe (Lightfield 1997). In a pest-specific risk assessment, Fowler and Lakin (2001) concluded that if this pest if it were introduced it would be highly likely to encounter a suitable climate and hosts in much of the US and cause severe economic and environmental consequences.

A brief history of the direct effects of *S. littoralis* on the quantity and quality of food and fiber crops is provided by USDA (1982) and suggests that the pest can have devastating consequences. In the late 1930s, *S. littoralis* lowered cotton yields by as much as 75% (reviewed in USDA 1982). Currently, this pest remains “one of the most destructive agricultural lepidopterous pests within its subtropical and tropical range. It can attack numerous economically important crops all the year round” (reviewed in CABI/EPPO 1997). The pest is particularly problematic on vegetables, ornamentals, and leguminous forage in the Mediterranean (reviewed in Inserra and Calabretta 1985, reviewed in CABI/EPPO 1997) and on cotton in Egypt (reviewed in Inserra and Calabretta 1985, reviewed in CABI/EPPO 1997).

The economic consequences of establishment by *S. littoralis* would not be limited to its direct effects on production agriculture; *S. littoralis* could also adversely affect access to foreign markets. The European and Mediterranean Plant Protection Organization (EPPO) considers *S. littoralis* an A2 quarantine pest; the pest also has quarantine status with the Caribbean Plant Protection Commission (CPPC), the Organismo Internacional Regional de Sanidad Agropecuaria (OIRSA), the North American Plant Protection Organization (NAPPO), Belarus, Russia, Ukraine, and Turkey (EPPO 1999).

Symptoms

Larvae feed on leaves, stems, fruit, or pods of plants in any growth stage (CAB 2000). As larvae feed on the underside of leaf surfaces, they skeletonize leaves and create leaf scars (reviewed in USDA 1982). Feeding can occur anywhere on a leaf. Feeding will eventually produce holes or “bare sections” on “leaves, young stalks, bolls, and buds” (reviewed in USDA 1982). Later instars may bore into stems. Damage to the stem may cause a plant to wilt distal to the entry hole. Damage may occur from spring to fall (reviewed in USDA 1982).

Known Hosts

Spodoptera littoralis has hosts in 40 plant families. Host plants have been reviewed in CAB (2000), Brown (1975), Salama (1971), Zhang (1994), and Pogue (2003).

Hosts	**References**
Abelmoschus esculentus (okra)	(Pogue 2003)
Acacia nilotica (gum arabic tree)	(Pogue 2003)
Alcea rosea (hollyhock)	(CAB 2000)
Allium cepa (onion)	(Pogue 2003)
Amaranthus retroflexus (redroot amaranth)	(Pogue 2003)
Anemone sp. (anemone)	(Pogue 2003)
Apium graveolens (wild celery)	(Pogue 2003)
Arachis hypogaea (peanut/groundnut)	(Pogue 2003)
Arum sp. (arum)	(Pogue 2003)
Beta vulgaris (beet/sugarbeet)	(Zhang 1994, Pogue 2003)
Boerhavia erecta (erect spiderling)	(Pogue 2003)
Brassica oleracea var. *gemmifera* (brussels sprouts)	(Pogue 2003)
Brassica rapa ssp. *rapa* (rape mustard)	(Salama et al. 1971, Pogue 2003)
Brassica spp. (cruciferous crops)	(Pogue 2003)
Caladium sp. (caladium)	(Pogue 2003)
Callistephus chinensis (China aster)	(Pogue 2003)
Camellia sinensis (tea)	(Pogue 2003)
Canna sp. (canna)	(Pogue 2003)
Capsicum sp., *C. annuum* (garden pepper)	(Salama et al. 1971, Zhang 1994, Pogue 2003)
Capsicum sp. (pepper)	(Zhang 1994, Pogue 2003)
Casuarina equisetifolia (beach sheoak)	(Salama et al. 1971, Pogue 2003)
Centrosema sp. (butterfly pea)	(Pogue 2003)
Chenopodium murale (nettleleaf goosefoot)	(Pogue 2003)
Chrysanthemum sp. (chrysanthemum)	(Zhang 1994, Pogue 2003)
Cichorium intybus (chicory)	(Pogue 2003)
Citrullus lanatus (melons)	(Pogue 2003)

Hosts	**References**
Citrus spp. (oranges/other citrus)	(Zhang 1994, Pogue 2003)
Citrus spp., *C. aurantium* (citrus/sour orange)	(Salama et al. 1971, Pogue 2003)
Coffea arabica (coffee)	(Pogue 2003)
Colocasia esculenta (coco yam)	(Pogue 2003)
Convolvulus sp., *C. arvensis* (bindweed/field bindweed)	(Pogue 2003)
Corchorus capsularis, *C. olitorius* (jute/nalta jute)	(Pogue 2003)
Cryptomeria sp. (Japanese cedar)	(Pogue 2003)
Cucumis melo (melons)	(Zhang 1994)
Cucurbita pepo (gourd/pumpkin)	(Zhang 1994, Pogue 2003)
Cupressus sp., *C. lusitanica* var. *lusitanica* (cypress)	(Pogue 2003)
Cynanchum sp., *C. acutum* (swallow-wort)	(Pogue 2003)
Cynara cardunculus (cardoon)	(Pogue 2003)
Dalbergia sissoo (Indian rosewood)	(Pogue 2003)
Datura sp. (datura)	(Pogue 2003)
Daucus carota ssp. *sativus* (carrot)	(Salama et al. 1971, Pogue 2003)
Dendranthema sp., *D. indicum* (daisy)	(Pogue 2003)
Dianthus barbatus (sweetwilliam)	(Pogue 2003)
Dianthus caryophyllus (carnation)	(Pogue 2003)
Dillenia indica (chulta)	(Pogue 2003)
Eleusine coracana (finger millet)	(Pogue 2003)
Eucalyptus globulus (Tasmanian bluegum)	(Salama et al. 1971, Pogue 2003)
Eucalyptus saligna (Sydney bluegum)	(Pogue 2003)
Euphorbia heterophylla (Mexican fireplant)	(Pogue 2003)
Ficus carica (fig/edible)	(Pogue 2003)
Ficus sp., *F. variegata* (fig)	(Salama et al. 1971, Pogue 2003)
Fragaria vesca (woodland strawberry)	(Pogue 2003)
Gerbera sp. (gerbera daisy)	(Pogue 2003)
Gladiolus sp. (gladiolus)	(Pogue 2003)
Glycine max (soybean)	(Zhang 1994, Pogue 2003)
Gnaphalium sp., *G. luteo-album* (Gnaphalium)	(Pogue 2003)
Gossypium hirsutum, *G. barbadense* (cotton)	(Salama et al. 1971, Zhang 1994, Pogue 2003)

Hosts	**References**
Helianthus annuus (sunflower)	(Zhang 1994, Pogue 2003)
Helianthus tuberosus (Jerusalem artichoke)	(Pogue 2003)
Heliotropium europaeum (European heliotrope)	(Pogue 2003)
Hibiscus cannabinus (brown indianhemp)	(Pogue 2003)
Hibiscus mutabilis (Dixie rosemallow)	(Pogue 2003)
Indigofera tinctoria (true indigo)	(Pogue 2003)
Ipomoea batatas (sweet potato)	(Salama et al. 1971, Zhang 1994, Pogue 2003)
Lactuca sativa (lettuce)	(Pogue 2003)
Lantana sp. (lantana)	(Pogue 2003)
Linum usitatissimum (flax)	(Pogue 2003)
Luffa aegyptiaca (sponge gourd)	(Pogue 2003)
Lycopersicon esculentum (tomato)	(Zhang 1994, Pogue 2003)
Macadamia ternifolia (macadamia nut)	(Pogue 2003)
Maesopsis eminii (umbrella-tree)	(Pogue 2003)
Malus domestica (apple)	(Pogue 2003)
Malva parviflora (cheeseweed mallow)	(Pogue 2003)
Malva sylvestris (high mallow)	(Pogue 2003)
Manihot esculenta (tapioca)	(Pogue 2003)
Medicago sativa (alfalfa/lucerne)	(Pogue 2003)
Mentha spicata (spearmint)	(Pogue 2003)
Monstera deliciosa (tarovine)	(Pogue 2003)
Morus sp. (mulberry)	(Salama et al. 1971, Pogue 2003)
Muehlenbeckia platycladum (ribbon bush)	(Pogue 2003)
Musa x *paradisiaca* (=*Musa paradisiaca*) (banana, plantain)	(Salama et al. 1971)
Nicandra physaloides (apple of Peru)	(Pogue 2003)
Nicotiana glauca (tree tobacco)	(Pogue 2003)
Nicotiana tabacum (tobacco)	(Zhang 1994, Pogue 2003)
Ocimum basilicum (sweet basil)	(Pogue 2003)
Opuntia sp., *Cactus opuntia* (pricklypear)	(Pogue 2003)
Oryza sativa (rice)	(Salama et al. 1971, Zhang 1994, Pogue 2003)
Pericallis x *hybrida* (florist's cineraria)	(Pogue 2003)
Persea americana (avocado)	(Pogue 2003)

Hosts	**References**
Petroselinum crispum (parsley)	(Pogue 2003)
Phaseolus vulgaris (bean/green bean/kidney bean/dry edible bean)	(Zhang 1994, Pogue 2003)
Philodendron sp., *P. domesticum* (philodendron)	(Pogue 2003)
Phoenix dactylifera (date palm)	(Salama et al. 1971, Pogue 2003)
Phytolacca dodecandra (pokeweed)	(Pogue 2003)
Pinus sp., *P. kesiya* (pine)	(Pogue 2003)
Piper sp. (pepper)	(Pogue 2003)
Pistia stratiotes (water lettuce)	(Pogue 2003)
Pisum sativum (pea)	(Pogue 2003)
Poaceae (grasses)	(CAB 2000)
Populus alba (white poplar)	(Pogue 2003)
Portulaca oleracea (little hogweed)	(Pogue 2003)
Prunus domestica (plum)	(Pogue 2003)
Psidium guajava (guava)	(Salama et al. 1971, Pogue 2003)
Psychotria sp., *P. capensis* (wild coffee)	(Pogue 2003)
Punica granatum (pomegranate)	(Salama et al. 1971, Pogue 2003)
Pyrus communis (pear)	(Pogue 2003)
Quercus petraea (durmast oak)	(CAB 2000)
Raphanus sativus (radish)	(Pogue 2003)
Rheum rhabarbarum, *R. rhaponticum* (rhubarb)	(Pogue 2003)
Ricinus communis (castor bean)	(Salama et al. 1971, Zhang 1994, Pogue 2003)
Rosa spp. (roses)	(Pogue 2003)
Rumex acetosa (sorrel)	(Pogue 2003)
Saccharum officinarum (sugarcane)	(Pogue 2003)
Salvia officinalis (sage)	(Pogue 2003)
Sesamum orientale (sesame)	(Pogue 2003)
Sesbania sesban (Egyptian riverhemp)	(Pogue 2003)
Solanum melongena (aubergine/eggplant)	(Salama et al. 1971, Zhang 1994, Pogue 2003)
Solanum sp., *S. anguivi*, *S. grandiflorum*, *S. villosum* (nightshade)	(Pogue 2003)
Solanum torvum (turkey berry)	(Pogue 2003)
Solanum tuberosum (potato)	(Salama et al. 1971, Zhang 1994, Pogue 2003)

Hosts	**References**
Sorghum bicolor (sorghum)	(Pogue 2003)
Spinacia oleracea (spinach)	(Pogue 2003)
Tectona grandis (teak)	(CAB 2000)
Theobroma cacao (cocoa/cacao)	(Pogue 2003)
Trifolium sp., *T. alexandrinum* (clover/Egyptian clover)	(Salama et al. 1971, Pogue 2003)
Trigonella foenum-graecum (sicklefruit fenugreek)	(Pogue 2003)
Triticum aestivum (wheat)	(Pogue 2003)
Verbena sp. (verbena/vervain)	(Pogue 2003)
Vicia faba (broad bean/horsebean)	(Pogue 2003)
Vigna unguiculata (blackeyed pea/cowpea)	(Pogue 2003)
Viola odorata (sweet violet)	(Pogue 2003)
Vitis vinifera (grape)	(Salama et al. 1971, Pogue 2003)
Xanthosoma mafaffa (giant golden taro)	(Pogue 2003)
Zea mays (corn)	(Salama et al. 1971, Zhang 1994, Pogue 2003)
Zinnia violacea (elegant zinnia)	(Pogue 2003)

Known Distribution

Spodoptera littoralis occurs in Africa, the Mediterranean, and the Middle East (reviewed in Zhang 1994).

Location	**References**
Algeria	(Brown and Dewhurst 1975)
Angola	(Brown and Dewhurst 1975)
Botswana	(Brown and Dewhurst 1975)
Burkina Faso	(Brown and Dewhurst 1975)
Burundi	(Brown and Dewhurst 1975)
Cameroon	(Brown and Dewhurst 1975)
Central African Republic	(Brown and Dewhurst 1975)
Chad	(Brown and Dewhurst 1975)
Comoros	(Brown and Dewhurst 1975)
Congo Democratic Republic	(Brown and Dewhurst 1975)
Egypt	(Brown and Dewhurst 1975, Zhang 1994)
Eritrea	(Brown and Dewhurst 1975)
Ethiopia	(Brown and Dewhurst 1975)
Gabon	(Brown and Dewhurst 1975)

Location	References
Gambia	(Brown and Dewhurst 1975)
Ghana	(Brown and Dewhurst 1975)
Greece	(Zhang 1994)
Guinea	(Brown and Dewhurst 1975)
Israel	(Zhang 1994)
Italy	(Zhang 1994)
Ivory Coast	(Brown and Dewhurst 1975)
Kenya	(Brown and Dewhurst 1975)
Liberia	(Brown and Dewhurst 1975)
Libya	(Brown and Dewhurst 1975)
Madagascar	(Brown and Dewhurst 1975)
Madeira	(Brown and Dewhurst 1975)
Malawi	(Brown and Dewhurst 1975)
Mali	(Brown and Dewhurst 1975)
Mauritania	(Brown and Dewhurst 1975)
Mauritius	(Brown and Dewhurst 1975)
Morocco	(Brown and Dewhurst 1975)
Mozambique	(Brown and Dewhurst 1975)
Niger	(Brown and Dewhurst 1975)
Nigeria	(Brown and Dewhurst 1975)
Portugal	(Zhang 1994)
Réunion	(Brown and Dewhurst 1975)
Rwanda	(Brown and Dewhurst 1975)
São Tomé and Príncipe	(Brown and Dewhurst 1975)
Saudi Arabia	(Brown and Dewhurst 1975)
Senegal	(Brown and Dewhurst 1975)
Seychelles	(Brown and Dewhurst 1975)
Sierra Leone	(Brown and Dewhurst 1975)
Spain	(Zhang 1994)
Somalia	(Brown and Dewhurst 1975)
South Africa	(Brown and Dewhurst 1975)
Sudan	(Brown and Dewhurst 1975)
Swaziland	(Brown and Dewhurst 1975)
Tanzania	(Brown and Dewhurst 1975)
Togo	(Brown and Dewhurst 1975)
Tunisia	(Brown and Dewhurst 1975)
Uganda	(Brown and Dewhurst 1975)
Yemen	(Brown and Dewhurst 1975)

Location	References
Zambia	(Brown and Dewhurst 1975)
Zimbabwe	(Brown and Dewhurst 1975)

Potential Distribution within the US

Spodoptera littoralis appears to survive in dry, tropical, or temperate climates (CAB 2000). Available geographic records of the distribution of *S. littoralis*, suggest the species may be most closely associated with deserts and xeric shrublands; Mediterranean scrub; temperate broadleaf and mixed forests; tropical and subtropical grasslands, savannas, and shrublands; and tropical and subtropical moist broadleaf forests. Based on the distribution of climate zones in the US, we estimate that approximately 49% of the continental US would be suitable for *S. littoralis*. This prediction generally concurs with the findings of Fowler and Lakin (2001) but suggests that the southeastern US may be less favorable, and the upper Midwest more favorable, than they had predicted.

Survey

A number of sampling considerations for *S. littoralis* have been proposed (reviewed in USDA 1982). Surveys for this pest can take place any time during the growing season while plants are actively growing. Early instars (<3rd) are likely to be on lower leaf surfaces during the day. Larvae will skeletonize leaves by feeding on this surface and subsequent damage to the leaf provides evidence of the presence of larvae. Sweep net sampling may be effective at dawn or dusk. Specimen identification should be confirmed by a trained taxonomist. However, not all sampling methods are equally effective for all life-stages of the insect. Eggs are only likely to be found by visual inspection of leaves. First through third instars may be detected by sweep net sampling; nearly all instars can be detected by visual inspection of plants; and, later instars (4th-6th) and pupae may be found by sieving soil samples (Abul-Nasr and Naguib 1968, Abul-Nasr et al. 1971).

Active traps (either light- or pheromone-based) have been recommended for monitoring relative densities of adults (DEFRA 1999). The synthetic sex pheromone (*Z*,*E*)-(9,11)-tetradecadienyl acetate has proven highly effective at trapping male moths of *S. littoralis* (Salem and Salama 1985). Sex-pheromone baited delta traps remained attractive for approximately 2 weeks, but effectiveness declined after 3 to 4 weeks of use (Ahmad 1988). To monitor male flight activity in vegetable production areas, delta traps were placed 1.7 m [~5 ½ ft] above the ground at a rate of 2 traps/ha [approximately 1 trap/acre] (Ahmad 1988). Pheromone lures impregnated with 2 mg of the pheromone blend (blend not specified) were replaced after 4 weeks of use (Ahmad 1988). Traps are deployed at a similar height (1.5 m) [~5 ft] to monitor male flight in cotton (Salem and Salama 1985). Catches in pheromone traps did not correlate as well with densities of egg-masses in cotton fields as did catches in a black-light trap (Rizk et al. 1990a). The attractiveness of traps baited with (*Z*,*E*)-(9,11)-tetradecadienyl acetate is governed primarily by minimum air temperature; relative humidity, adult abundance, wind velocity, densities of female *S. littoralis* also affect the number of males that are captures at different times of the year (Rizk et al. 1990b).

Lures for *S. littoralis* can be used in the same traps with lures for *S. litura*, *Helicoverpa armigera*, *Pectinophora scutigera* (all not known to occur in the US), and *P. gossypiella* (exotic established in US). Lures for *S. littoralis* may also attract *Erastria* sp. (established in US) (PPQ 1993).

Light traps using a 125 W mercury-vapor bulb have been used to non-discriminately capture multiple *Spodoptera* spp. (Blair 1974) and most assuredly other insects as well. A modified light trap using six 20-W fluorescent lights also proved an effective for monitoring flight activity of *S. littoralis* (El-Mezayyen et al. 1997).

Larvae of *S. littoralis* can be confused with *S. exigua* (established in the US), but *S. littoralis* larva are light or dark brown, while *S. exigua* are brown or green (Brown and Dewhurst 1975). *Spodoptera littoralis* is also larger than *S. exigua*. *Spodoptera littoralis* larvae can also be confused with *S. litura*, but *S. litura* larvae have bold lateral spots present from the mesothorax to the eighth abdominal segment. Neither *S. littoralis* nor *S. litura* are established in the US. Adults of these two *Spodoptera* spp are almost indistinguishable based on external color and pattern. See Pogue (2002) for a detailed list of characters used to separate the species. Identification of *S. littoralis* must be confirmed by a well trained entomologist.

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Thaumatotibia leucotreta[*]

Scientific Name

Thaumatotibia leucotreta Meyrick

Synonyms:
At the generic level:
Thaumatotibia Zacher, 1915: 529-Heppner, 1980: 34 (as synonym of *Cryptophlebia*). Type species: *Thaumatotibia roerigii* Zacher, 1915 [=*Argyroploce leucotreta* Meyrick, 1913] by monotypy

- *Argyroploce* Hübner, [1825]
- *Olethreutes* Hübner, 1822, Syst. -alphab. Verz.: 58-67, 69, 72. Type species: *Phalaena arcuella* Clerck, 1759, Icon. Insect. Rariorum 1: pl 10 fig. 8, by subsequent designation by Walsingham, 1895, Trans. Ent. Soc. Lond. 1895: 518.
- *Metriophlebia* Diakonoff, 1969: 89. –Razowski, 1977: 259. –Clarke, 1986: 162 (as synonym of *Cryptophlebia*), syn. n. Type species: *Eucosoma chaomorpha* Meyrick, 1929, by monotypy

At the species level:
leucotreta (Meyrick) 1913. Ann. Transv. Mus. 3: 267-336.
roerigii Zacher, 1915: 529 Beiträge zur Kenntnis der westafrikanischen Planzenschädlinge.-Tropenpflanzer 18: 504-534.

Common Names

False codling moth

Type of Pest

Moth, fruit/seed feeder

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera,
Family: Tortricidae

Reason for inclusion in manual

CAPS Priority Pest (FY 2007)

Pest Description

“Small to medium-sized, grayish-brown to dark brown/black moths with broad forewings (forewing index: 0.41-0.44 mm in males, and 0.38-0.42 mm in females)

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[*] This document is largely excerpted from the report: Venette, R.C., E.E. Davis, M. DaCosta, H. Heisler, and M. Larson. 2003. Mini-Risk Assessment: False Codling Moth, *Thaumatotibia* (=*Cryptophlebia*) *leucotreta* (Meyrick) [Lepidoptera: Tortricidae]. Available on-line at www.aphis.usda.gov/ppq/ep/pestdetection/pra/tleucotretapra.pdf

with a blackish triangular pretornal patch. Externally species of *Thaumatotibia* are similar to species of *Cryptophlebia*. Wing venation of *Thaumatotibia* is characterized by a small accessory cell delineated by the chorda from between R2 and R3 (closer to R3) to R4 or from between R1 and R2 (very close to R2) to between R5 and R5 (Fig. 4, description of venation), or the absence of accessory cell (the chorda coincident with the margin of the discal cell), and by a short discal cell in the hindwing, especially in the male (0.42-0.43x length of the wing). Eighth tergum in male with a broadly sclerotized plate with convex posterior margin and laterally produced into curved points, with paired patches of long mane-like scales, but without a pair of long filiform scale tufts from shallow membranous pockets on each side of eighth tergum as in *Cryptophlebia*. Male genitalia are characteristic in the large, ovate valva (the outer surface with a patch of very long, curled scales, which is shared with *Cryptophlebia*), in the sacculus often with teeth distally, and in the juxta sometimes producing caudally a pair of denticulate, ovate lobes (the *chaomorpha*-group). The female genitalia is characterized by the sterigma indicated by an ovate or rectangular sclerite, connecting posteriorly with a pair of ovate granulations with modified scales, by the corpus bursae with a ring of granulation at the juncture of the ductus bursae, and sometimes a diverticulum ventrally or laterally" (Fig. 1) (Komai 1999).

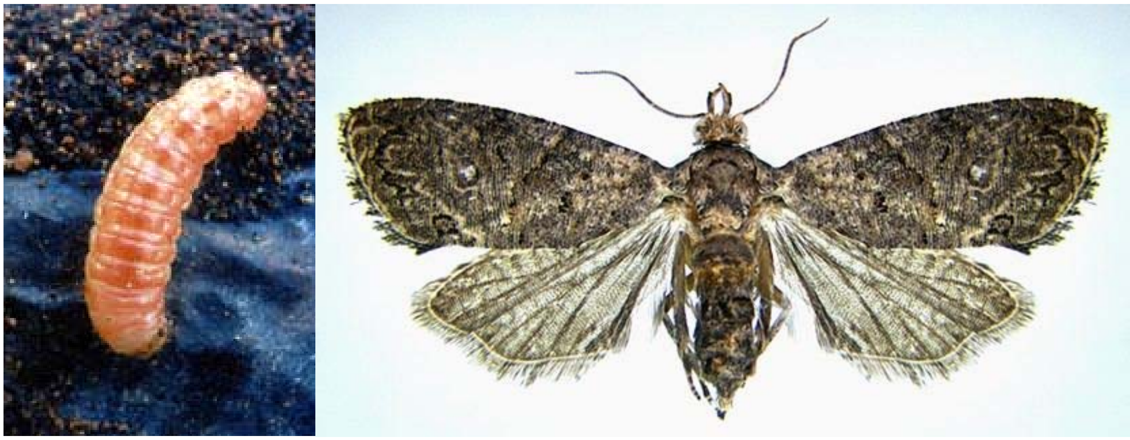


Fig. 1. Larva and adult of *T. leucotreta*. Images not to scale.
[Larval image from http://www.arc.agric.za/institutes/ itsc/main/avocado/moth.htm; Adult image from Georg Goergen/IITA Insect Museum, Cotonou, Benin as published in (CAB 2003)]

Head: "Frons with very dense, erect and moderately long scales. Antenna filiform, less than 2/3 length of forewing. Labial palps long and wavy; second segment widened distally, but scales appressed and rather short; terminal segment extends forward horizontally, about 1/3 length of second, slender, with appressed scales, apex blunt" (Fig. 2) (Komai 1999).

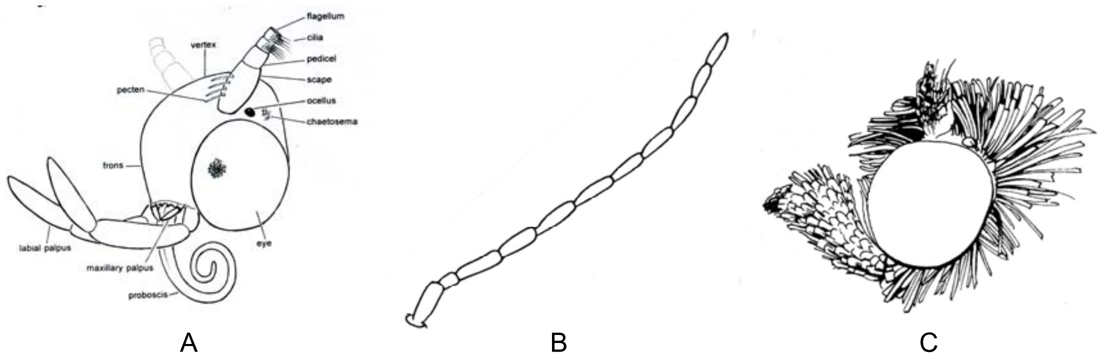


Fig. 2. Lateral views of head: A-Ventrolateral view of general moth head; B-Filiform antenna; and C-Lateral view head of *Thaumatotibia hemitoma* (Diakonoff)-male.
[Images reproduced from A-Robinson et al. (1994); B-Borror et al. (1989); C- Komai (1999)]

Thorax: “Posterior crest present. Hind tibia with modified scales on inner side, the inner apical spur enlarged with a batch of scales, the bases of which have a layer of secreting cells” (Fig. 3) Komai (1999).

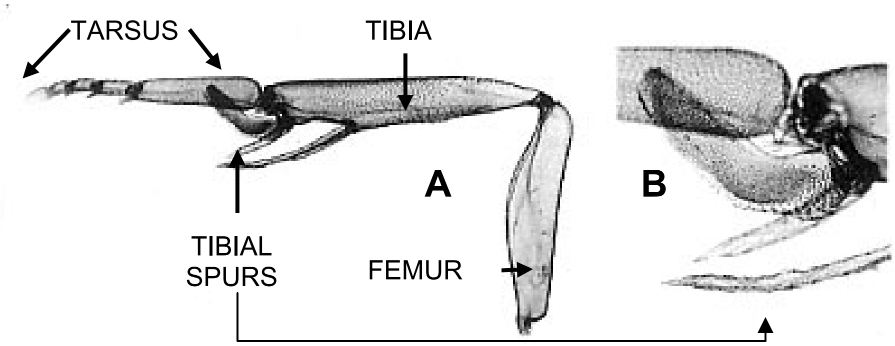
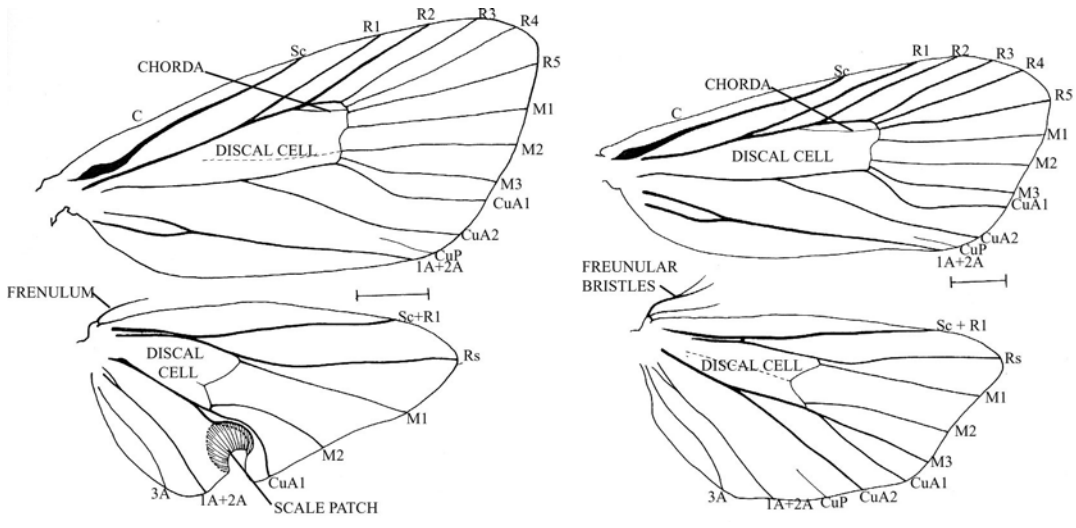


Fig. 3. Photomicrograph of hind tibia of *Thaumatotibia* sp.: A-Morphology of hindtibia with modified scales and apical spur removed. B-Detail of tibial spurs.
[Image reproduced from Komai (1999)]

Wings: “Forewing pattern a mixture of bluish-gray, brown, black, and rust colored red-brown markings, the most conspicuous is the blackish triangular pre-tornal marking and the crescent-shaped marking above it, and a minute white spot in the discal area” (Bradley et al. 1979).

Venation: “There is a scent organ on the distal 2/3 of CuA2 on upper side. Its presence is indicated by concavity on wing membrane bounded with thickened ridges bearing the secreting cells” (reviewed in Komai 1999).



A B

Fig. 4. Venation of *Thaumatotibia leucotreta* (Meyrick), A-male, B-Female.
Veins: A-anal; C-Costa, Cu-Cubitus (CuA1-1st anterior cubitus; CuA2-2nd anterior cubitus; CuP-posterior cubitus); D-discal cell; M-Media, R-Radius, Sc-Subcosta.
[Reproduced from Komai (1999).]

Larva: "Body length of mature larva 15 mm. Head yellowish-brown. Body orange or pink in final instar. Pinacula large, darker than body color. Spiracle on A8 near the posterior margin. Prolegs with 31-40 crochets arranged in a biordinal circle. Anal fork present. Chaetotaxy SD1 and SD2 on same pinaculum on A9; SV group on A1-A6 trisetose, on A7 and A8 bisetose, A9 unisetose; L group trisetose on A9" (Fig. 5) (Komai 1999).

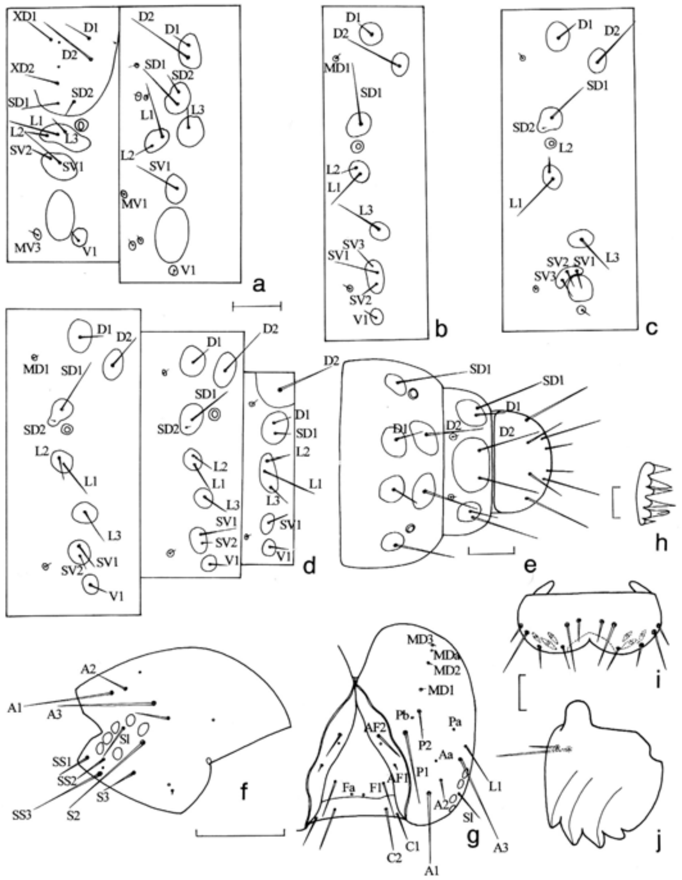


Fig. 5. Setal map of *Thaumatotibia leucotreta*; drawing scale a-g: 0.5 mm, h-j: 0.1 mm
[Reproduced from Komai (1999)]

Pupa: "Body length 6-10 mm. Body pale yellowish-brown. Similar to *Cryptophlebia*. Spiracles transversely ovate. A2-A7 with two rows of dorsal spines; A8-A10 with one row of strong spines, in male A8 with two rows of dorsal spines; A10 with a pair of strong spines along anal rise, without hooked setae except two pairs along anal rise" (Fig. 6) (Komai 1999).

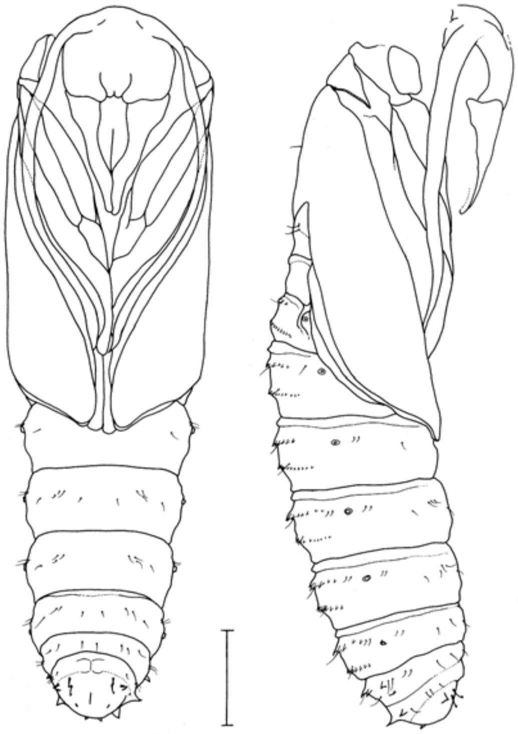


Fig. 6. Pupa of *Thaumatotibia sp*.: left, ventral view; right, lateral view (scale = 1 mm) [Reproduced from Komai (1999)]

Biology and Ecology

Thaumatotibia leucotreta has 2-10 generations annually (Daiber 1980, Couilloud 1994, Begemann and Schoeman 1999). If food is available and climate is appropriate, *T. leucotreta* can remain active throughout the year (Blomefield 1978, Newton 1988a). In South Africa, moths may live 1-6 weeks, or up to ~28 weeks under favorable winter conditions (Daiber 1980, Couilloud 1994). At warmer temperatures (e.g., 20-25°C [68-77°F]) the life span of moths is shorter, while at cooler temperatures (e.g., 10-15°C [50-59°F]), reproduction is generally greater (Daiber 1980). On average, females live longer than males (Daiber 1980). The ratio of males to females is 1:2 (Couilloud 1994).

Emergence occurs early in the morning (Couilloud 1994). Moths are active at night and spend daytime hours resting on shaded portions of the host (Blomefield 1978, Couilloud 1994). Moth activity increases with the onset of host flowering (Newton 1989b). Moths can mate several times per day (Couilloud 1994). Oviposition occurs on or near developing fruit after petal fall (Daiber 1975, Newton 1989a, Ochou 1993).

Females tend to choose smooth, non-pubescent surfaces for egg-laying. On cotton, green bolls are preferred (Couilloud 1994). On peach, eggs are deposited near fruit on smooth leaves (Blomefield 1978, Newton 1988a). Moths also tend to select areas on fruit with damage (Blomefield 1978, Newton and Crause 1990).

Oviposition begins 2-3 days after females emerge (Blomefield 1978). A female will generally produce between 87-456 eggs depending on temperature (within a range of 15-25°C [59-77°F]) (Daiber 1980). However, individual female fecundity can vary from 5-799 eggs (Daiber 1980). Eggs are laid singly or in small groupings of 2-4 “overlapping like tiles” (Daiber 1980, Blomefield 1989, Newton and Crause 1990, Couilloud 1994). Eggs are only laid in the evening between 5 and 10 pm (Daiber 1980).

Egg development takes 2-22 days depending on temperature (Daiber 1979). Eggs are extremely sensitive to cold temperatures and extended periods of low humidity. Temperatures below 0°C [<32°F] over a 2-3 day period can kill eggs (Blomefield 1978, Daiber 1979).

Thaumatotibia leucotreta has up to 5 instars (Bradley et al. 1979, Couilloud 1994). Larvae may be present from 4-173 days, depending on temperature and host plant (Blomefield 1978; Daiber 1979b}(Daiber 1979, Daiber 1989, Couilloud 1994). The last instar is typically completed in fruit if it is available. A larva prepares to pupate by leaving the fruit and spinning a cocoon with silk and soil particles (reviewed in USDA 1984). Pupation occurs on the soil surface, in the soil, in crevices under bark, in dropped fruit or in debris (Blomefield 1978, USDA 1984, La Croix and Thindwa 1986a, Daiber 1989, Newton and Crause 1990).

Pupae emerge slightly from the cocoon before adult emergence takes place. “The empty pupal skin usually remains attached to the cocoon” (Daiber 1989). Under laboratory conditions, the pupal stage lasts between 2-33 days, depending on temperature (Daiber 1989). Pupae are also sensitive to cold temperatures and heavy rainfall (Daiber 1989). Pupae that have completed ¼ to ½ of their development tend to be more cold resistant than older or younger pupae (Myburgh and Bass 1969).

Several studies have described the developmental threshold and degree days necessary for the completion of each life stage (Table 1).

Table 1. Developmental threshold and degree day requirements for *Thaumatotibia leucotreta*

Stage	Developmental threshold (˚C)	Degree Days (± SE)	Notes	Reference
Egg	11.93	51.2-69.3	Lab study	(Daiber 1979)
	11.7	69.4±3.2	Calculated from author’s data	(Daiber 1975)

Stage	Developmental threshold (˚C)	Degree Days (± SE)	Notes	Reference
Larva	11.6-12.5	156	Lab study	(Daiber 1979)
Pupa	11.9	174 (females) 186 (males)	Lab study	(Daiber 1979)
Adult	8	232±4.2	Male life span; calculated from author's Table 1	(Daiber 1980)
	8.1	229.8±3.8	Male life span; calculated from author's Table 5	(Daiber 1975)
	9.5	243.5±12.3	Female life span; calculated from author's Table 1	(Daiber 1980)
	9.7	237.8±10.9	Female life span; calculated from author's Table 5	(Daiber 1975)
	6.4	242.8±18.1	Oviposition period; calculated from author's Table 5	(Daiber 1975)
	12.2	79.2±3.8	Time to 50% eggs laid; calculated from author's Table 1	(Daiber 1980)
	15	12.8±2.8	Preoviposition period; calculated from author's Table 5	(Daiber 1975)

Pest Importance

Thaumatotibia leucotreta is a significant pest of fruit trees and field crops in portions of Africa (reviewed in Zhang 1994). *Thaumatotibia. leucotreta* is a pest of economic importance to several crops, including: corn, cotton, citrus, lychee, macadamia, peach and plum, throughout sub-Saharan Africa, South Africa, and the islands of the Atlantic and Indian Oceans (Schwartz and Kok 1976, Daiber 1979, 1980, La Croix and Thindwa 1986a, 1986b, Wysoki 1986, Blomefield 1989, Newton 1989a, Newton and Crause 1990, Silvie 1993, Sétamou et al. 1995). Damage to corn is caused from larvae entering the ear from the husk through the silk channel (Ndemah et al. 2001).

Damage caused by this insect on oak is not well described. The larva is able to bore into acorns where it feeds (Anderson 1986). The incidence of acorn infestation varies seasonally from ca. 1-20% with greatest infestation occurring in late summer. Acorn may provide an important "bridge" resource to sustain the population when other fleshy fruits are not available.

All stages of citrus and stone fruits are vulnerable to attack (Newton 1988b). *Thaumatotibia leucotreta* larvae are capable of developing in hard green fruit before control measures can be started (Catling and Aschenborn 1974). Once a fruit is damaged, it becomes vulnerable to fungal organisms and scavengers (Newton 1989b). In peaches, up to 28% loss of late-peach crops has been reported (reviewed in CAB 2003). Larvae damage stone fruits as they burrow into the fruit at the stem end and begin to feed around the stone (Blomefield 1978). On oranges, *T. leucotreta* caused 2-5% damage on Valencia and Navel oranges in 1954 (reviewed in USDA 1984), but yield losses have been as great as 10-20% (reviewed in CAB 2003).

Thaumatotibia leucotreta has caused significant yield losses (≥30%) to macadamia crops in Israel and South Africa (La Croix and Thindwa 1986a, Wysoki 1986). Damage to macadamia nuts is caused from larvae feeding on the developing kernel after they pierce the husk and shell (La Croix and Thindwa 1986a). Nuts reaching 14 – 19 mm diameter size are at the most risk because nutrient content is the highest. *T. leucotreta* reaches the adult stage by this point and is able to oviposit on these nuts (La Croix and Thindwa 1986a).

In Ugandan cotton, *T. leucotreta* caused 20% loss of early sown varieties and 42 - 90% loss of late varieties (Byaruhanga 1977). Larval penetration of cotton bolls facilitates entry of other microorganisms that can rot and destroy the boll (Couilloud 1994).

Symptoms

Thaumatotibia leucotreta is known primarily as a borer, not a defoliator, and symptoms caused by its feeding are best described for agricultural commodities. In general, *T. leucotreta* prefers to feed within the fruit of a host. Larval feeding and development can affect fruit development at any stage, causing premature ripening and fruit drop (Schwartz and Kok 1976, USDA 1984, Newton 1988b, 1989b, Begemann and Schoeman 1999). Infestation of soft fruits can be identified by the brown spots and dark brown frass (Blomefield 1978). Detecting infested peaches can be difficult if fruit is still firm and abscission has not occurred (reviewed in USDA 1984). An infested orange will show brown, sunken spots surrounding holes where larvae have entered (Bradley et al. 1979).

Evidence of feeding in oaks may be similar to that of macadamia. In macadamia, larvae enter a nut through the side of the shell before the shell is fully hardened (La Croix and Thindwa 1986a). Aside from the entry hole and perhaps a hollowed nut, no other symptoms are likely to indicate the presence of the insect in oak.

Known Hosts

False codling moth feeds on more than 70 host plants (CAB 2003).

Hosts	**References**
Abelmoschus esculentus (okra)	(USDA 1984; CAB 2003)
Abutilon hybridum (Indian mallow)	(CAB 2003)
Abutilon spp. (jute)	(USDA 1984)
Ananas comosus (pineapple)	(USDA 1984; CAB 2003)
Annona muricata (soursop)	(USDA 1984; CAB 2003)
Annona reticulata (custard apple)	(USDA 1984)
Averrhoa carambola (carambola)	(USDA 1984; CAB 2003)
Bauhinia galpini (Pride of De Kaap)	(Anon. 1983)
Calotropis procera (sodom apple)	(USDA 1984)
Camellia sinensis (tea)	(USDA 1984; CAB 2003)
Capparis tomentosa (wig-‘n-bietjie)	(USDA 1984)
Capsicum spp. (pepper/pimento)	(Bourdouxhe 1982, USDA 1984, CAB 2003)
Catha edulis (khat)	(USDA 1984)
Ceiba pentranda (kapok/copal)	(USDA 1984; CAB 2003)
Chrysophyllum palismontatum (stamvrugte)	(Anon. 1983)
Citrus sinensis, *Citrus* spp. (citrus)	(Schwartz 1979; Anon. 1983; USDA 1984; Daiber 1989; Newton 1989; Zhang 1994; CAB 2003)
Coffea arabica, *Coffea spp*. (coffee)	(USDA 1984; CAB 2003)
Cola nitida (cola)	(USDA 1984)
Combretum apiculatum (rooibos/bushwillow)	(Anon. 1983; USDA 1984)
Combretum zeyheri (raasblaar)	(USDA 1984)
Diospyros mespiliformis (jakkalsbessie)	(USDA 1984)
Diospyros spp. (persimmon)	(Anon. 1983; USDA 1984)
Eugenia uniflora (Surinam cherry)	(USDA 1984)
Ficus capensis (fig, wild)	(USDA 1984)

Hosts	**References**
Garcinia mangostana (mangosteen)	(USDA 1984)
Gossypium hirsutum, *Gossypium* spp. (cotton)	(Reed 1974, USDA 1984, Silvie 1993, Zhang 1994, CAB 2003)
Harpephyllum caffum (kaffir plum)	(USDA 1984)
Hibiscus spp. (mallow)	(USDA 1984)
Juglans regia (English walnut)	(USDA 1984)
Litchi chinensis (lychee)	(USDA 1984; CAB 2003)
Macadamia ternifolia (macadamia nut)	(La Croix 1986; Zhang 1994; CAB 2003)
Mangifera indica (mango)	(USDA 1984, Javai 1986, CAB 2003)
Musa paradisiaca (banana)	(USDA 1984)
Olea europaea subsp. *europaea* (olive)	(USDA 1984; CAB 2003)
Pennisetum purpureum (elephant grass)	(USDA 1984)
Persea americana (avocado)	(USDA 1984; CAB 2003)
Phaseolus lunatus (lima bean)	(USDA 1984)
Phaseolus spp. (bean)	(USDA 1984)
Podocarpus falcatus (yellow-wood berries)	(Anon. 1983; USDA 1984)
Prunus persica (peach)	(Daiber 1980; Anon. 1983; USDA 1984; Daiber 1989; Zhang 1994; CAB 2003)
Prunus spp. (stone fruit)	(Anon. 1983; USDA 1984)
Pseudolachnostylis maprouneifolia (kudu-berry)	(USDA 1984)
Psidium guajava (guava)	(Anon. 1983; USDA 1984; CAB 2003)
Punica granatum (pomegranate)	(Anon. 1983; USDA 1984; CAB 2003)
Quercus spp. (oak)	(USDA 1984; Zhang 1994; CAB 2003)
Ricinus communis (castorbean)	(Del Valle and March 1972, USDA 1984, CAB 2003)
Royena pallens (bloubos)	(Anon. 1983; USDA 1984)
Schotia afra (boerboon)	(Anon. 1983; USDA 1984)
Sclerocarya caffra, S.birrea (marula)	(Anon. 1983)
Sida spp. (sida)	(USDA 1984)
Sorghum spp. (sorghum)	(USDA 1984; Zhang 1994; CAB 2003)
Syzygium cordatum (water-bessie)	(USDA 1984)
Theobroma cacao (cacao)	(USDA 1984)
Triumfeta spp. (bur weed)	(USDA 1984)
Vangueria infausta (wild medlar)	(Anon. 1983; USDA 1984)

Hosts	**References**
Vigna unguiculata, *Vigna* spp. (cowpea)	(USDA 1984)
Vitis spp. (grape)	(Zhang 1994)
Ximenia caffra (suurpruim/large sour plum)	(Anon. 1983; USDA 1984)
Zea mays (corn)	(USDA 1984; Zhang 1994; Sétamou 1995; CAB 2003)
Zizyphus jujuba (jujube)	(USDA 1984)
Zizyphus mucronata (buffalo thorn)	(USDA 1984)

Known Distribution

Thaumatotibia leucotreta is native to the Ethiopian zoogeographic province and presently occurs in much of Sub-Saharan Africa (reviewed in CAB 2003)

Location	**References**
Angola	(CIE 1976; CAB 2003)
Benin	(CIE 1976; CAB 2003)
Burkina Faso	(CAB 2003)
Burundi	(CIE 1976; CAB 2003)
Cameroon	(CIE 1976; CAB 2003)
Central African Republic	(CAB 2003)
Chad	(CIE 1976; CAB 2003)
Congo Democratic Republic	(CIE 1976; CAB 2003)
Ethiopia	(CIE 1976; CAB 2003)
Gambia	(CIE 1976; CAB 2003)
Ghana	(CIE 1976; CAB 2003)
Ivory Coast	(CIE 1976; CAB 2003)
Israel	(CAB 2003)
Kenya	(CIE 1976; CAB 2003)
Madagascar	(CIE 1976; CAB 2003)
Malawi	(CIE 1976; CAB 2003)
Mali	(CIE 1976; CAB 2003)
Mauritius	(CIE 1976; CAB 2003)
Mozambique	(CIE 1976; CAB 2003)
Niger	(CIE 1976; CAB 2003)
Nigeria	(CIE 1976; CAB 2003)
Rwanda	(CIE 1976; CAB 2003)
Réunion	(CIE 1976; CAB 2003)
Saint Helena	(CIE 1976; CAB 2003)
Senegal	(CIE 1976; CAB 2003)
Sierra Leone	(CIE 1976; CAB 2003)

Location	References
Somalia	(CIE 1976; CAB 2003)
South Africa	(CIE 1976; CAB 2003)
Sudan	(CIE 1976; CAB 2003)
Swaziland	(CAB 2003)
Tanzania	(CIE 1976; CAB 2003)
Togo	(CIE 1976; CAB 2003)
Uganda	(CIE 1976; CAB 2003)
Zambia	(CAB 2003)
Zimbabwe	(CIE 1976; CAB 2003)

Potential Distribution within the US

The currently reported global distribution of *T. leucotreta* suggests that the pest may be most closely associated with biomes that are generally classified as desert and xeric shrubland; tropical and subtropical grasslands, savannas, and shrubland; and tropical and subtropical moist broadleaf forests. Based on the distribution of climate zones in the US, we estimate that approximately 20% of the continental US may be suitable for *T. leucotreta*.

Our analysis is generally consistent with the speculation of Karvonen (1983), who suggested that this species was only likely to survive in “hot tropical or subtropical areas.” The predicted absence of *T. leucotreta* from much of California concurs with Daiber (1989), who suggests that this pest may not perform well in Mediterranean climates, as found in portions of South Africa. The analysis differs somewhat from the suggestion that the pest may be able to establish in areas where the average annual low temperature is >-10°C [14°F] (PPQ 1993).

Survey

Visual inspections of plant materials may be used to detect eggs, larvae, and adults of *T. leucotreta* (reviewed in USDA 1984). Eggs will commonly be found on fruits, foliage, and occasionally on branches (reviewed in USDA 1984). On citrus fruits and other fleshy hosts, dissections are needed to detect larvae; larvae are likely to be found in the pulp (reviewed in USDA 1984). Infested fruits may be on or off the tree. In cotton, older larvae may be found in open bolls and cotton seed (reviewed in USDA 1984). Occasionally adults may be observed on the trunk and leaves of trees in infested orchards (reviewed in USDA 1984). For field crops such as corn, the whole plant is the recommended sample unit (Schulthess et al. 1991). Because larvae of *T. leucotreta* have a strongly aggregated spatial distribution among corn plants, a large sample size (>60 plants) is recommended (Schulthess et al. 1991, Ndemah et al. 2001); however, at low densities of the pest (<1 larva/plant) sample sizes may be prohibitively large to detect the pest (Schulthess et al. 1991).

Robinson black light traps are ineffective at attracting adult *T. leucotreta* (Begemann and Schoeman 1999). Therefore, black light traps should not be used. This recommendation stands in stark contrast to the experience of Reed (1974) who used Robinson black light traps to monitor adult *T. leucotreta* in cotton for nearly 4 years. The effectiveness of black light traps may be improved if used in conjunction with pheromone lures (Möhr 1973). Möhr (1973) speculates that pheromone may provide a long-distant attractant, but that attraction to black light becomes much stronger when moths are in close proximity to light traps.

Sex pheromones have been identified, and the synthetic compounds are highly attractive to males of *T. leucotreta*. Males are attracted to a two component blend of (*E*)-8-dodecenyl acetate and (*Z*)-8-dodecenyl acetate (Persoons et al. 1976, 1977, Newton et al. 1993). These components are most effective when used in a ratio between 70:30 and 30:70 (*E*:*Z*) (Persoons et al. 1976, 1977, Angelini 1979, Angelini et al. 1981, Bourdouxhe 1982). More recently, Newton et al. (1993) refined the sex pheromone and reported that a 90:10 ratio was optimal. A loading rate between 0.5 and 1.0 mg per septum was found to attract the greatest number of males (Jactel and Vaissayre 1988). The pheromone blend (1 mg applied to a rubber septum) has been used effectively with Pherocon 1C traps to capture male *T. leucotreta* (Newton et al. 1993). Delta traps have also been used (Newton 1988a, 1989a, Newton and Mastro 1989, La Croix 1990), but these have performed less well than either the Hoechst Biotrap or Pherocon 1C traps (Newton and Mastro 1989, Ochou 1993). Traps using closed polyethylene vials to dispense pheromones captured more moths than traps using rubber septa (using a 50:50 blend of (E)- and (Z)-8-dodecenyl acetate La Croix et al. 1985). Lures should be replaced every 2-4 weeks (Daiber 1978, Jactel and Vaissayre 1988)

Traps should be placed approximately 1.5m [5 ft] high (Blomefield 1989, Newton and Mastro 1989, Newton et al. 1993). Lures should be replaced every 8 wks (PPQ 1993). For routine monitoring, 2-5 traps/ha [1-2 traps/a] is recommended (http://www.insectscience.co.za/phertraps.htm). Pheromone traps (homemade design with unspecified pheromone blend) have been used to monitor the number of *T. leucotreta* adult males in citrus orchards (Daiber 1978) and detect the presence of the pest in peach orchards (Daiber 1981).

Lures for *T. leucotreta* should not be used in the same trap with lures for the pink bollworm (*Pectinophora gossypiella*) because the combination of lures results in fewer pink bollworm captures (Schwalbe and Mastro 1988). Lures for *T. leucotreta* can be used in the same trap with lures for *P. scutigera* (Schwalbe and Mastro 1988).

Pheromone lures with (*E*)- and (*Z*)-8-dodecenyl acetate may also attract *Cydia cupressana* (native), *Hyperstrotia* spp. (PPQ 1993), *Cydia atlantica* (exotic)

(Chambon and Frerot 1985), *Cydia phaulomorpha* (exotic) and *Cryptophlebia peltastica* (exotic) (Bourdouxhe 1982, Newton et al. 1993).

Thaumatotibia leucotreta can be confused with many *Cydia* spp. including *C. pomonella* (codling moth) because of similar appearance and damage, however, unlike codling moth its host range does not include apples, pears or quince (reviewed in USDA 1984). *Cydia pomonella* (Figs. 7-10) is well established in the US. "In West Africa, *T. leucotreta* is often found in conjunction with *Mussidia nigrevenella*," however they can be distinguished by close examination of morphological characters (CAB 2003). In South Africa, there is also an overlapping host range for *T. leucotreta* and *Cydia peltastica*, particularly on lychee and macadamia (Newton and Crause 1990).

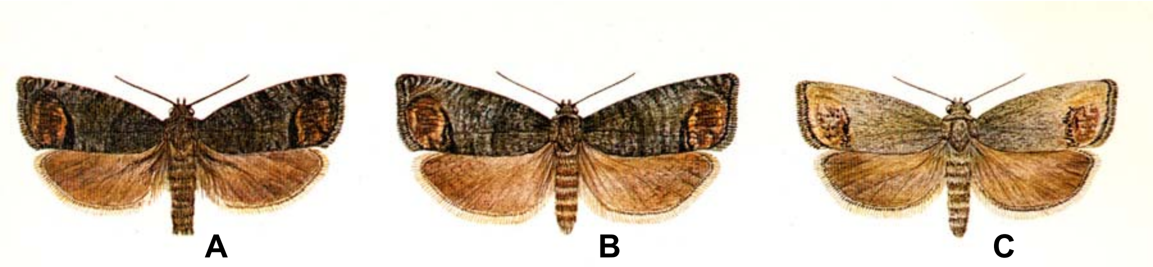


Fig. 7. Dorsal views of *Cydia pomonella* (Linnaeus) A-male, B & C-female (to illustrate degree of morphological variation within a sex).
[Reproduced from Bradley et al. (1979)]

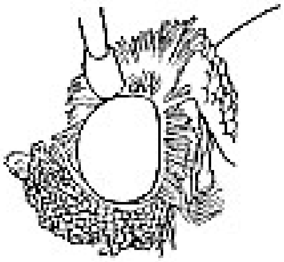


Fig. 8. Lateral view of head of *Cydia pomonella* (Linneaus)
[Reproduced from Bradley et al. (1979)]

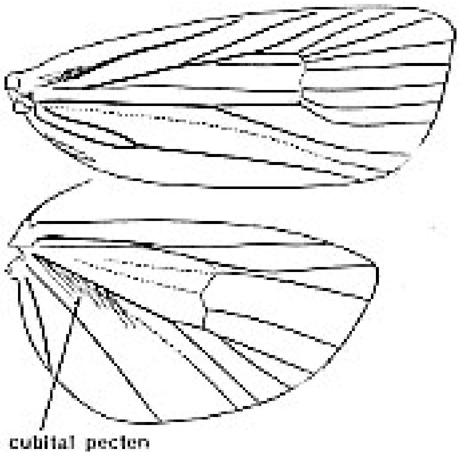


Fig. 9. Venation of *Cydia pomonella*
[Reproduced from Bradley et al. (1979)]

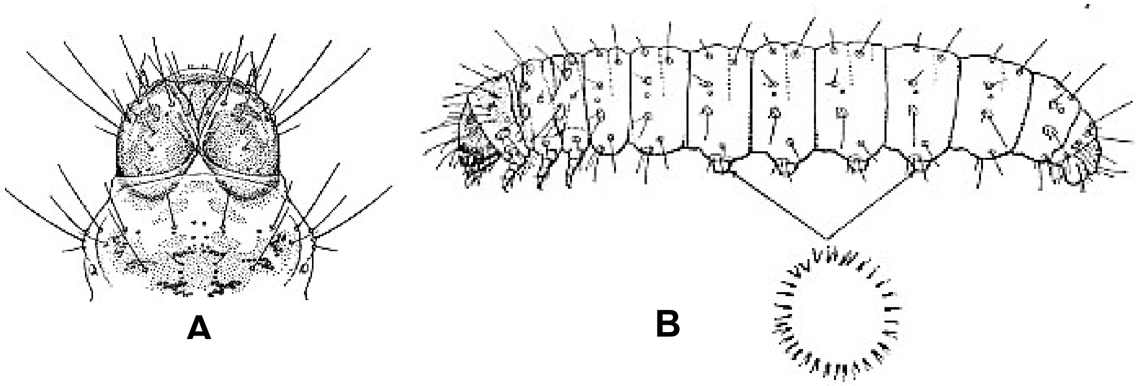


Fig. 10. Larva of *Cydia pomonella* (Linneaus). A-Dorsal view of head, B-lateral view of body; pattern of crochets
[Reproduced from Bradley et al. (1979)]

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Tortrix viridana

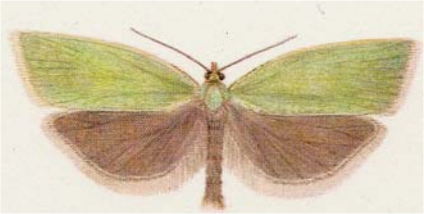

Fig. 1. Drawing of male *Tortrix viridana*.
[Image reproduced from Bradley et al. (1973)]

Scientific Name

Tortrix viridana (Linnaeus)

Synonyms:
Phalaena viridana Linnaeus
Heterognomon viridana Barrett 1905
Tortrix viridana Pierce & Metcalfe 1922

Common Names

green oak tortrix
oak leaf roller
oak roller moth

Type of Pest

Moth, leafroller, defoliator

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda, **Order:** Lepidoptera, **Family:** Tortricidae, **Subfamily:** Tortricinae

Reason for inclusion in manual

Exotic Forest Pest Information System – classified as a very high risk pest with the potential to attack oaks

Pest Description

Males and females with a wingspan of “17-24 mm; males under 17 mm are known, the smallest being an individual from Oxshott (Surrey) [UK] with a wing span barely 13 mm, but such undersized specimens are rare. Sexual dimorphism not pronounced; antenna of male weakly dentate, densely ciliate, cilia less than width of flagellum, posterior margin of flagellum thinly clad with whitish scales; female usually with abdomen stouter and wings broader, antenna filiform, sparsely ciliate, more densely clad with whitish scales” (Bradley et al. 1973).

“Male. Forewing light green (viridine), darker basally, delicately strigulate distally, head, thorax, patagium and tegula concolorous, vertex and patagium often tinged with yellow; labial palpus whitish, suffused with fuscous exteriorly; whole of costal margin of forewing narrowly edged with whitish yellow, base of costa sometimes suffused with fuscous; dorsal scale-tuft tinged with yellow; cilia whitish, a pale green sub-basal line. Hindwing light grey; cilia whitish, with a grey sub-basal line around apex and along termen” (Fig. 1) (Bradley et al. 1973).

“Female. Similar to male” (Fig. 2) (Bradley et al. 1973).

“Variation. This species shows little variation except in the depth and intensity of the green coloration of the forewing and in the strength of the strigulation. Specimens are sometimes found in which the green coloration is replaced by dull yellow or primrose yellow; these apparently represent a recurring genetical form and are not due to abrasion or discoloration” (Bradley et al. 1973).

Fig. 2. Male (right) and female (left) of *Tortrix viridana* on an oak leaf.
[Image from Louis-Michel Nageleisen, #UGA2101020, www.insectimages.org]

Egg. “Pale yellow at first, becoming orange-brown later, lenticular, delicately sculptured, usually covered by green scales from the upper surface of the forewings” (Bradley et al. 1973).

“Larva. Head shining blackish brown or black; prothoracic plate varying from light greenish brown to green or grey, sometimes mottled with brown, a narrow whitish medial sulcus, posterior margin edged with black, anterior margin sometimes with a whitish border, the coloration and markings being exceedingly variable (in early instar larvae the prothoracic plate is usually entirely black); abdomen light olive-green, greyish green in early instars, integument strongly shagreened; pinacula dark brown or black; anal plate dark brown or green; anal comb usually with 8 prongs; thoracic legs shining black” (Fig. 3) (Bradley et al. 1973).

Fig. 3. Larva of Tortrix viridana on outer surface of rolled leaf
[Image from Milan Zubrick, #1370047, www.invasive.org]

"Pupa. Varying from brown to black" (Fig. 4) (Bradley et al. 1973).

Fig. 4. Pupa of *Tortrix viridana* within folded oak leaf.
[Image from Petr Kapitola, #UGA2112047, www.insectimages.org]

Biology and Ecology

Tortrix viridana typically has one generation per year, but two are possible in warmer climates (Bogenschütz 1991). Moths are active from late April to early July in Europe and Eurasia. The life span for adults is approximately one week. Moths are active during the day and may be found in large numbers in the upper portion of tree crowns. Mating occurs during the afternoon and evening; however, the species may be able to reproduce parthenogenetically (Du Merle 1999a). Adults are capable of utilizing winds to disperse up to 100 km [~62 mi] (Du Merle and Pinguet 1981, Schneider 1984, Winter 1984, Bogenschütz 1991, reviewed in Ciesla 2003). The sex ratio for *T. viridana* is 1:1. One female may deposit 50-60 eggs, usually in clutches of 2-3 eggs that partially overlap (reviewed in Horstmann 1977, Bogenschütz 1991, Markov 1993). Egg masses are deposited in a lustrous, sticky substance on bark, leaf scars or near buds on small shoots (2 year old growth; 1-3 cm diameter) (Speight 1985, Bogenschütz 1991, Markov 1993).

Fecundity and development of *T. viridana* are affected by food (quantity and quality) and temperature (Bogenschütz 1991, Markov 1993). Overcrowding or reduced food quality results in fewer, smaller adults and reduced oviposition rates. Because males require less food than females during larval development, they pupate and emerge earlier in the year (Bogenschütz 1991). When food is scarce, females are more likely than males to die from starvation, which alters the sex ratio (Bogenschütz 1991).

Eggs enter diapause and overwinter (Fazeli and Abai 1990). Diapause begins in the spring or early summer and ends in late fall-early winter (Du Merle 1999b). Temperatures must drop to at least 8°C [~46°F] for eggs to satisfy cold requirements. Although the effect of days colder than 8°C on the duration of diapause has not been quantified, diapause will end relatively sooner in areas with colder climates (Du Merle 1999b). Eggs must experience 60-100 days at 8°C [~46°F] to break diapause (Du Merle 1999b). Once diapause has broken, embryos within eggs will begin to develop once temperatures reach about 10°C [50°F] and will hatch when 200 degree days [°C] have accumulated (reviewed in Bogenschütz 1991).

Larvae hatch in the spring of the year after eggs were laid (Du Merle 1982, Speight 1985, Ivashov et al. 2002). Hatch occurs near the time of budbreak (Speight 1985, Ivashov et al. 2002), typically in April or early May (Bradley et al. 1973). Newly hatched larvae feed on opening buds, young leaves, flowers and new shoots (Speight 1985, reviewed in Ciesla 2003). Larvae develop through five instars, and insects remain as larvae for 20-40 days depending on temperature (Speight 1985, Fazeli and Abai 1990, Tiberi and Roversi 1990, Bogenschütz 1991, reviewed in Ciesla 2003, reviewed in CAB 2006). Early instars are vulnerable to cold temperatures. A spring freeze with temperatures at -6°C [21°F] will kill nearly 30% of the population (reviewed in Bogenschütz 1991). When food becomes scarce, larvae will hang from a silken thread and disperse by ballooning (reviewed in CAB 2006). If hatching does not coincide with budbreak, larvae will survive for about 10 days under spring conditions. If food cannot be found in that time, larvae will die from starvation (Speight 1985, Hunter 1990, Bogenschütz 1991, Markov 1993) or simply fail to develop as a result of reduced nutritional quality of leaves or host plant resistance mechanism (protease inhibitor) (Hunter 1990, 1997, Ivashov et al. 2001, 2002). For these reasons, early hatched larvae are more likely to complete development than those that hatch later in spring (Ivashov et al. 2001).

Pupation occurs in rolled leaves spun together with silk. The duration of the pupal stage generally lasts 2-3 weeks, depending on temperature (Bogenschütz 1991, reviewed in CAB 2006).

See ‘Known Hosts’ for a listing of the plants that can be attacked by *T. viridana* and ‘Pest Importance’ for a discussion of the potential economic impact.

Pest Importance

Tortrix viridana is considered a pest of economic importance in Europe, Russia and the Near East where periodic outbreaks have contributed to extensive oak defoliation and decline (Fazeli and Abai 1990, Hunter 1990, Tiberi and Roversi 1990, Markov 1993). In Spain, this pest has been a significant pest of oak; larval feeding damage to tender bark and new shoots has resulted in reduced flowering and fruiting (acorn production) (reviewed in Ciesla 2003). In Poland, *T. viridana* and other defoliators have impacted over 650 thousand ha [>1½ million acres] and prompted control measures (Stocki 1994). *Tortrix viridana* can cause serious damage to oak forests, particularly when egg hatch occurs early in spring and conditions for larval development are optimal (Ivashov et al. 2001). Larval density may vary from 1-2 per bud, and up to 12 per bud in heavy infestations. The final two larval instars are the most damaging (reviewed in CAB 2006).

Larvae are considered largely monophagous feeders of oaks, particularly new foliage and buds; however, this pest may feed on a variety of ornamental trees and shrubs (see 'Known Hosts') (Bogenschütz 1991, Du Merle 1999b). Population size can vary from year to year (see 'Biology and Ecology'), but in its native range, the insect is attacked by several natural enemies which may hold populations in check (reviewed in Ellis 1946, reviewed in Horstmann 1977, Hunter et al. 1997). Thus, the potential economic impact in the US in the absence of natural enemies is uncertain.

The economic impact of *T. viridana* is difficult to measure especially because this pest typically occurs with other primary defoliators (including *Operophthera brumata* and *Erannis defoliaria*) and secondary pests (Speight 1985, reviewed in Ciesla 2003, reviewed in CAB 2006). Considerable defoliation can occur without causing significant damage (Mattson and Addy 1975, Furniss and Carolin 1977, Drooz 1985). However, heavy defoliation repeated over a period years can cause decline of tree health, reduced growth rates, attraction of secondary pests, and tree death (Furniss and Carolin 1977, Drooz 1985, Speight 1985, Rubtsov 1996).

Tortrix viridana is not known to occur in the US. Risks associated with *T. viridana* have been evaluated for North American forests. The insect was considered of high risk, but this rating was very uncertain (Ciesla 2003). The potential for environmental impact was considered high, but the potential for establishment, spread and economic-impact were each rated moderate.

Symptoms

Tortrix viridana larvae feed on buds, foliage, flowers, and new shoot growth of oaks and other deciduous trees and shrubs. The insect may cause significant defoliation (see 'Pest Importance'). Developing larvae will roll leaves to create protected feeding sites and to pupate (see 'Biology and Ecology').

Fig. 5. Foliar feeding damage caused by *Tortrix viridana*.
[Image from Petr Kapitola, www.insectimages.org]

Feeding damage by *T. viridana* larvae is similar to that caused by other polyphagous defoliators. Evidence of the pest includes heavy defoliation in oak crowns; rolled leaves of host trees spun with silk webbing; feeding damage on buds, foliage and flowers of host trees.

If larvae feed on developing buds, the buds will assume a conical shape; webbing from bud to bud will be present, and a blackened exit hole (Bogenschütz 1991). The entire crown may be covered with webbing during severe outbreaks.

If infestations last several years, trunks will be shorter, bent and tapered. Epicormic shoots may form (Bogenschütz 1991).

Known Hosts

Larvae of *T. viridana* are reported predominantly as feeders of oak foliage, but may also feed on other tree and woody plant species. *Tortrix viridana* has a preference for *Quercus robur* and *Q. pubescens* (Novotný et al. 1990).

Hosts	**References**
Acer sp. (maple)	(Zhang 1994, Meijerman and Ulenberg 2000, Ciesla 2003, CAB 2006)
Acer monspessulanum (Montpellier maple)	(CAB 2006)
Carpinus sp. (hornbeam)	(Zhang 1994, Meijerman and Ulenberg 2000, Ciesla 2003)
Carpinus betulus (hornbeam)	(CAB 2006)
Corylus avellana (hazel)	(CAB 2006)
Fagus sp. (beech)	(Zhang 1994, Meijerman and Ulenberg 2000)
Fagus sylvatica (beech)	(Ciesla 2003)
Fraxinus angustifolia (narrow-leaved ash)	(CAB 2006)
Juniperus sp. (juniper)	(CAB 2006)
Picea abies (common spruce)	(CAB 2006)
Populus sp. (poplar)	(Zhang 1994, Meijerman and Ulenberg 2000, Ciesla 2003)
Quercus spp. (oak)	(Ellis 1946, Ortiz and Templado 1976, Speight 1985, Tiberi and Roversi 1990, Van der Geest and Evenhuis 1991, Markov 1993, Zhang 1994, Du Merle 1999b, Meijerman and Ulenberg 2000, Matosevic 2001)
Quercus canariensis (Algerian oak)	(CAB 2006)
Quercus cerris (European turkey oak)	(Zhang 1994, CAB 2006)
Quercus faginea (Lusitanian oak)	(CAB 2006)
Quercus frainetto (Hungarian oak)	(CAB 2006)
Quercus iberica	(CAB 2006)

Hosts	**References**
Quercus ilex (holm oak)	(Du Merle and Pinguet 1981, Du Merle 1982, Van der Geest and Evenhuis 1991, Du Merle 1999b, Ciesla 2003, CAB 2006)
Quercus imeretina	(CAB 2006)
Quercus petraea (durmast oak)	(Horstmann 1977, Hunter 1990, Van der Geest and Evenhuis 1991, Stocki 1994, Ciesla 2003, CAB 2006)
Quercus pubescens (downy oak)	(Du Merle and Pinguet 1981, Du Merle 1982, Novotný et al. 1990, Du Merle 1999b, CAB 2006)
Quercus pyrenaica (black oak)	(Van der Geest and Evenhuis 1991, CAB 2006)
Quercus robur (common oak)[1]	(Horstmann 1977, Winter 1984, Hunter and Willmer 1989, Hunter 1990, Novotný et al. 1990, Van der Geest and Evenhuis 1991, Stocki 1994, Rubtsov 1996, Hunter et al. 1997, Ciesla 2003, CAB 2006)
Quercus rubra (northern red oak)	(Van der Geest and Evenhuis 1991, Stocki 1994)
Quercus sessiliflora	(Du Merle 1999b)
Quercus suber (cork oak)	(Van der Geest and Evenhuis 1991, Du Merle 1999b, Ciesla 2003, CAB 2006)
Quercus trojana (Macedonian oak)	(CAB 2006)
Rhododendron sp.	(Zhang 1994)
Rubus idaeus (raspberry)	(Meijerman and Ulenberg 2000, CAB 2006)
Salix sp. (willow)	(Zhang 1994, Meijerman and Ulenberg 2000)
Urtica sp. (nettle)	(Meijerman and Ulenberg 2000)
Vaccinium sp. (blueberry)	(Zhang 1994, Meijerman and Ulenberg 2000)

1. Rubtsov (1996) recognized two “phenoforms”: late oaks (*Quercus robur* var. *tardiflora*) and early oaks (*Quercus robur* var. *praecox*)

Known Distribution

Tortrix viridana is reported from:

Location	**References**
Algeria	(Ciesla 2003)
Azerbaijan	(Meijerman and Ulenberg 2000)
Croatia	(Matosevic 2001)
Cyprus	(Ciesla 2003)

Location	References
France	(Du Merle and Pinguet 1981, Du Merle 1982, 1999b, CAB 2006)
Germany	(Arn et al. 1979, Knauf et al. 1979, Schneider 1984)
Iran	(Fazeli and Abai 1990, Zhang 1994)
Israel	(Ciesla 2003)
Italy	(Tiberi and Roversi 1990, CAB 2006)
Macedonia	(CAB 2006)
Morocco	(Ciesla 2003, CAB 2006)
Poland	(Stocki 1994)
Portugal	(CAB 2006)
Romania	(Zhang 1994)
Russia	(Markov 1993, Rubtsov 1996)
Scandinavia	(Ortiz and Templado 1976)
Slovakia	(Novotný et al. 1990)
Spain	(Ortiz and Templado 1976, CAB 2006)
Syria	(Ciesla 2003)
Tunisia	(Ciesla 2003)
Turkey	(Zhang 1994)
United Kingdom	(Winter 1984, Speight 1985, Hunter and Willmer 1989, Hunter 1990, Hunter et al. 1997, Ciesla 2003)

Potential Distribution within the US

Based on where *T. viridana* has been reported worldwide, we predict that the species is most closely associated with biomes characterized as temperate coniferous forest or Mediterranean scrub. Collectively these two biomes account for 20% of the area within the contiguous US. The biomes occur along the coastal west; Pacific northwest, higher elevations in the intermountain west, and coastal southeast. A specialized environment will be required for establishment of this insect because cold temperatures are of unique importance. The environment must provide an adequate number of chilling days for the insect to complete diapause; however, springs must be reliably warm to prevent young larvae from dying due to a spring freeze.

Survey

Visual sampling can be used to detect populations of *T. viridana,* but this method is not generally recommended. Severe infestations can cause extensive defoliation and stunting of trees (see 'Symptoms' above). However, many of these symptoms can be caused by other oak defoliators. Other established tortricids may also roll leaves. Infestations of *T. viridana* can be difficult to confirm because larvae tend to occupy the upper canopy (Bogenschütz 1991). Visual sampling for eggs is difficult and subject to errors.

Pheromones have been identified for *T. viridana* (El-Sayed 2005). Pheromone-baited traps are extremely useful to detect the presence of the species, but may have limited utility to accurately estimate population abundance (reviewed in CAB 2006). The primary component of the sex pheromone for *T. viridana* is (Z)-11-tetradecenyl acetate, which is an effective single-compound attractant for the species (Arn et al. 1979, Knauf et al. 1979). The pheromone for the red banded leaf roller, *Argyrotaenia velutinana*, is comprised of a 2:3 ratio of (Z)-11-tetradecenyl acetate and dodecyl acetate, which attracts *T. viridana* (Hrdý et al. 1979). This blend is available commercially. Novotný (1990) noted that (Z)-11-tetradecenyl acetate in a 9:1 ratio with (Z)-11-tertadecnol attracted two to five times as many males as (Z)-11-tetradecenyl acetate alone.

The pheromone is effective with several trap types (Novotný et al. 1990), Pherocon 1C traps are common (Hrdý et al. 1979). Trap color has no affect on the number of captured males (Schneider 1984). Traps should be attached to the trunk of a tree or hung from a branch, placing the trap at eye level, approximately 1.5 m [5 ft] above ground (Bogenschütz 1991). Pheromone should be dispensed from rubber septa loaded with 1 mg of attractant (Hrdý et al. 1979). Traps should be separated by at least 50 m [ca. 160 ft] (Hrdý et al. 1979).

Traps baited with pheromone for *T. viridana* (presumably (Z)-11-tetradecenyl acetate though not specified) may also attract: *Ptycholoma lecheana*, *Archips xylosteana*, *Archips crataegana*, *Aleimma loeflingiana*, *Choristoneura hebenstreitella*, *Croesia bergmanniana* (Tiberi and Roversi 1990).

Traps with the pheromone blend for red banded leaf roller may also attract *Aphelia paleana*, *Archips xylosteana*, *Argyrotaenia pulchellana*, and *Eupoecilia angustana* (Hrdý et al. 1979).

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Arthropods
Woodwasps

Tremex fuscicornis

Scientific Name

Tremex fuscicornis (Fabricius)

Synonyms:
Sirex fuscicornis Fabricius (Latreille)
Sirex camelogigas Christ
Sirex struthiocamelus Villers
Tremex juxicernis Walker
Urocerus fuscicornis Latreille
Xyloecematium fuscicornis Heyden
Xyloterus fuscicornis Boie
Tremex simulacrum Takeuchi

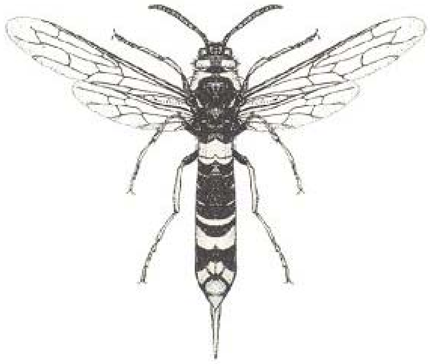

Fig. 1. Line drawing of *Tremex fuscicornis*
[Image from CSIRO, www.ento.csiro.au/aicn/sysstem/c_2267.htm]

Common Names

Tremex wasp

Type of Pest

Woodwasp, phloem feeder

Taxonomic Position

Kingdom: Animalia, **Phylum:** Arthropoda **Order:** Hymenoptera,
Family: Siricidae, **Subfamily:** Tremecinae

Reason for inclusion in manual

Exotic Forest Pest Information System – classified as a very high risk pest with the potential to attack oaks

Pest Description

Tremex fuscicornis is morphologically similar to *T. columba* and other siricids that occur in North America (Benson 1943). Closely related genera and species may be easily confused. Smith and Schiff (2002) provide a key to the genera of siricids of eastern North America. In Chile, where *T. fuscicornis* is exotic, the insect is most commonly confused with *Urocerus gigas* (Baldini 2001). In general, the genus *Tremex* is distinguishable from other siricids based on six morphological characters. Members of *Tremex* have (a) one apical spur on the hind tibia; (b) 14-15 segments per antenna; (c) no genal carina (i.e., a ridge in a space on the head behind the eyes; Fig. 2); (d) an ovipositor that is shorter than the length of the forewing; (e) an anal cell in the hindwing; and (f) a body without long golden hairs (Smith and Schiff 2002). Coloration is one of the characteristics used to distinguish woodwasp species, but colors vary considerably within a species, especially among males (Smith and Schiff 2002).

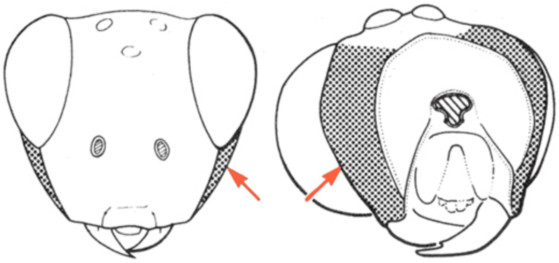

Fig. 2. Generalized line drawing of the gena, shaded areas indicated by arrows
[Image from www.hymatol.org/glossary/Pictures/occiput.jpg]

Benson (1943) distinguished between *T. fuscicornis* and *T. columba* by comparing three ratios which he considered to be diagnostic: 1) the ratio of the distance between the posterior ocelli (simple eyes) (POL) and the distance between a compound eye and the nearest posterior ocellus (OOL), 2) the ratio of the length of the ovipositor (measured from second valvifer or oblong plate base) to the length of the forewing (measured from tegula apex), and 3) the ratio of the length of the sawsheath, the apical portion of the ovipositor sheath, to the length of the ovipositor (measured as previously described). *Tremex fuscicornis* has a POL:OOL ratio of 1.2 (Benson 1943). The average length (to the nearest 0.5 mm) of the ovipositor is 18 mm, of the forewing is 21.5 mm, and of the sawsheath (from the apical tip of the ovipositor to the basal plate) is 7.5 mm. Consequently, the ovipositor:forewing ratio is 0.84, and the sawsheath:ovipositor ratio is 0.42 (Benson 1943). According to Benson (1943), these ratios provide more reliable taxonomic characters for the separation of species than the length of the ovipositor, ovipositor sections, or abdominal segments alone; these three characters are highly variable and can change as a result of desiccation. The values reported here reflect corrections to the original values reported by Benson (1943) who appears to have reported the inverse of each ratio. Witmond (1999) noted the error for the POL:OOL ratio.

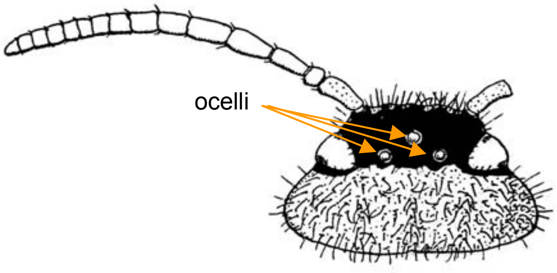

Fig. 3. Head of *Tremex fuscicornis*
[Image reproduced from Benson (1943)]

The ocelli are farther apart for *T. fuscicornis* (POL:OOL is ca. 2) than for *T. columba* (POL:OOL < 1.5) (Benson 1943). We encourage caution with the use of this character. Witmond (1999) noted that the reported ratios of POL:OOL did not match the verbal description of the distance between the ocelli. We presume Benson’s verbal account is correct but that the ratios were in error; consequently, we have corrected the ratios to match his verbal descriptions and line drawings.

Witmond (1999) suggests that postocellar distance is unreliable to separate species and recommends other characters such as the length of the setae (on head) and characteristics of the second recurrent vein (wing). Witmond (1999) provides the following description of a single female specimen: "... measures 23 mm from head apex of cornus.... The distances between the posterior ocelli (POL), between the anterior and a posterior ocellus (OL), between an eye and the nearest posterior ocellus (OOL), the diameter of the anterior ocellus, the length and width of the posterior ocelli are in the proportion of 25:11:10:10:13:10 [Fig. 3]). The antennae are 6 mm long, slightly swollen in the middle, with 14 segments. The forewing is 17 mm long. ... Brown setae cover head, thorax, trochanters and femora. ... The specimen is rather dark apparently, as both the head, except for a brown patch on the gena, and the thorax are black .... Antennae black, but segments 3, 4, and 14 more or less brown. The coxae, trochanters and femora are, with the exception of the distally brownish front femora, entirely black. All tibiae are yellow proximally, and brownish distally, the tarsi are either yellow or brownish. The first abdominal segment is entirely black, all the other segments black and yellow [Fig. 4]." Other authors have noted that a maximum body length of 40 mm and that the head and thorax are rust-colored (Witmond 1999).

Fig. 4. Female (left) and male (right) *Tremex fuscicornis*, not to scale.
[Images from: P. Parra S, www.infor.cl/webinfor/publicaciones/Documentos_2005/presentacion_silvotecnia.pdf]

Current species within *Tremex* have been described by Cameron (1889), Smith (1978), and Lee et al. (1998). *T. fuscicornis* has also been described by Precupetu and Negru (1961), and Lee et al. (1998).

Biology and Ecology

The biology of *T. fuscicornis* is poorly described, primarily because this pest is often concealed inside a host plant (reviewed in Drooz 1985, Smith and Schiff 2002, reviewed in Ciesla 2003). For this reason, it is difficult to specify a life span for this pest. The closely related *T. columba* may require 2 or more years from the time an egg is laid to complete development and begin to reproduce (Drooz 1985, Smith and Schiff 2002, reviewed in CAB 2006). In Chile, the number of generations per year is not known; however, larvae emerge in the fall from eggs laid in the summer (Gallegos Céspedes 2005).

As with other siricids, members of the genus *Tremex* are associated with basidiomycetous wood decay fungi (Drooz 1985, Smith and Schiff 2002). In North America, *Tremex columba* (see 'Pest Description') is associated with *Cerrena* (=*Daedalea*) *unicolor* (Drooz 1985, Smith and Schiff 2002). The same fungus was isolated from *Tremex longicollis* (Tabata and Abe 1995). In this symbiotic relationship, the fungus is carried to new tree hosts in specialized mycangia of the adult female, and then deposited under the bark or cambial layer when the female inserts her ovipositor into the tree to lay eggs. The fungus breaks down cellulose in the tree host with digestive enzymes and provides a source of nutrition for developing larvae. Whether the developing siricids actually feed on the digested wood products or solely on the fungus is not well understood (Morgan 1968, Smith and Schiff 2002). The larvae progressively tunnel into the wood of the host as they feed and develop over a period of 1-3 years (Smith and Schiff 2002). Pupation occurs close to the bark surface (Baldini 2001). Adult woodwasps emerge through circular shaped exit holes during summer and fall (reviewed in Drooz 1985, Smith and Schiff 2002, reviewed in Ciesla 2003). In Region V and the Área Metropolitana within Chile, adults emerge from October to January (spring-summer in South America) with maximum emergence occurring in the second and third week of November (Parra Sanhueza 2005). Adults have been observed into the fall (May) (Gallegos Céspedes 2005). Following a period of heavy attacks on poplar in Chile, it was estimated that a single poplar could produce 2000 adults (reviewed in Ciesla 2003). The male:female sex ratio is slightly female-biased as females comprise 55% of the population (Parra Sanhueza 2005).

See 'Known Hosts' for a listing of the plants that can be attacked by *T. fuscicornis* and 'Pest Importance' for a discussion of its potential economic impact.

Pest Importance

Like other siricids, *Tremex fuscicornis* is known to attack dead or declining trees but may also attack apparently healthy trees (Smith 1978, reviewed in Ciesla 2003). The economic impact of *T. fuscicornis* is difficult to measure especially because it can occur with other primary and secondary pests (Drooz 1985, Smith and Schiff 2002). However, in Chile, the insect seems to be particularly damaging to trees belonging to the genera *Salix* and *Populus* (Parra Sanhueza 2005). *Salix* is less affected than *Populus*, but the cumulative economic impacts are significant (reviewed in NAPPO 2006). The damage caused by symbiotic wood decay fungi may be more important than the damage caused solely by the insect. Fungi vectored by siricids are pathogenic; economic losses result from tree death, reduced growth, and reduced quality (Morgan 1968, Manion 1991, reviewed in NAPPO 2006). According to Smith (1978), *Tremex* spp. are generally not considered economically significant. Within its native range, *T fuscicornis* has not been reported as a pest; however, it has become a significant pest in new areas where it was accidentally introduced.

Risks associated with the introduction of *T. fuscicornis* into North America have been evaluated previously. Ciesla (2003) considered the potential for establishment, spread, and economic harm to be high. As a result, the overall, relative degree of risk was considered very high, but this assessment was very uncertain because the ability of this insect to compete with other established siricids in North America was (and remains) unknown (Ciesla 2003).

Symptoms

Symptoms of infection by *T. fuscicornis* and associated fungi are similar to those caused by other siricids: branch and crown dieback; reduced growth; yellowing leaves; wilted leaves; leaf and trunk necroses; tyloses formation; loosened bark; sapwood discoloration; and structural weakening (Drooz 1985, Smith and Schiff 2002, reviewed in Ciesla 2003).

Fig. 5. Exit holes produced as adults emerge from the main stem of a tree.
[Images from: P. Parra S, www.infor.cl/webinfor/publicaciones/Documentos_2005/presentacion_silvotecnia.pdf]

The only strong evidence for the presence of *T. fuscicornis* is the presence of exit holes (5-6 mm diameter) in the trunk (Gallegos Céspedes 2005). It is generally not possible to detect infestation in trees that were only recently attacked (Baldini 2001). Occasionally, a portion of the female ovipositor will remain in a tree (Baldini 2001).

Fig. 6. Detached ovipositor protruding from a tree (left) and close up (right). [Images from: P. Parra S, www.infor.cl/webinfor/publicaciones/Documentos_2005/presentacion_silvotecnia.pdf]

Known Hosts

Tremex fuscicornis attacks a wide range of deciduous tree species:

Hosts	**References**
Acer platanoides (Norway maple)	(Smith 1978, Lee et al. 1998)
Acer negundo (boxelder)	(Smith 1978, Lee et al. 1998)
Alnus japonica (Japanese alder)	(Smith 1978, Lee et al. 1998)
Alnus sp. (alder)	(Smith 1978, Lee et al. 1998)
Betula sp. (birch)	(Precupetu and Negru 1961, Smith 1978, Lee et al. 1998)
Betula pendula (common silver birch)	(CAB 2006)
Betula pubescens (downy birch)	(CAB 2006)
Carpinus betulus (European hornbeam)	(Smith 1978, Lee et al. 1998)
Celtis sinensis (Chinese hackberry)	(Smith 1978, Lee et al. 1998)
Fagus sylvatica (=F. "*silvatica*") (European beech)	(Precupetu and Negru 1961, Smith 1978, Lee et al. 1998)
Fagus sp. (beech)	(Smith 1978, Lee et al. 1998)
Juglans regia (English walnut)	(Smith 1978, Lee et al. 1998)
Juglans sinensis (Chinese walnut)	(Lee et al. 1998)
Populus alba (silver leaf or white poplar)	(CAB 2006)
Populus deltoides (Eastern cottonwood)	(CAB 2006)
Populus nigra (=*P. pyramidalis, P. nigra* var. *italica*) (Lombardy or black poplar)	(Precupetu and Negru 1961, Smith 1978, Lee et al. 1998)
Populus tremula (European aspen)	(Smith 1978, Lee et al. 1998)
Populus sp. (aspen/poplar)	(Precupetu and Negru 1961, Lee et al. 1998)
Prunus serrulata var. *spontanea* (Japanese flowering cherry)	(Lee et al. 1998)
Prunus ×yedoensis (pro sp.) (*subhirtella × speciosa*) (=*P. yedoensis*?) (chokecherry)	(Smith 1978, Lee et al. 1998)
Prunus sp.	(Smith 1978, Lee et al. 1998)
Pterocarya stenoptera (Chinese wingnut)	(Smith 1978, Lee et al. 1998)
Quercus sp. (oak)	(Smith 1978, Lee et al. 1998)
Robinia pseudoacacia (black locust)	(Smith 1978, Lee et al. 1998)
Salix sp. (willow)	(Smith 1978, Lee et al. 1998)
Salix babylonica (weeping willow)	(CAB 2006)

Hosts	References
Salix humboldtiana (Humboldt's willow)	(CAB 2006)
Ulmus davidiana var. *japonica* (=*U. japonica, U. propinqua*) (Japanese elm)	(Smith 1978, Lee et al. 1998)
Ulmus sp. (elm)	(Smith 1978, Lee et al. 1998)
Zelkova serrata (Japanese Zelkova)	(Smith 1978, Lee et al. 1998)
Zelkova sp.	(Smith 1978, Lee et al. 1998)

Known Distribution

Tremex fuscicornis has been reported from:

Location	References
Australia	(CSIRO 2004, CAB 2006)
Austria	(Smith 1978)
Bulgaria	(Stoyanov and Ljubomirov 2000)
Chile	(CAB 2006)
China (incl. Taiwan)	(Precupetu and Negru 1961, Smith 1978, Ciesla 2003)
Czechoslovakia	(Smith 1978)
Denmark	(Smith 1978)
Finland	(Smith 1978)
France	(Smith 1978, Witmond 1999)
Germany	(Smith 1978, Witmond 1999)
Hungary	(Smith 1978)
Italy	(Smith 1978)
Japan	(Precupetu and Negru 1961, Smith 1978, Lee et al. 1998)
Korea	(Precupetu and Negru 1961, Smith 1978, Lee et al. 1998)
Netherlands	(Witmond 1999)
Norway	(Smith 1978, Midtgaard et al. 1994)
Poland	(Smith 1978)
Romania (Rumanian People's Republic)	(Precupetu and Negru 1961, Smith 1978)
Russia (former USSR, incl. Asian and European Russia)	(Precupetu and Negru 1961, Lee et al. 1998, Ciesla 2003)
Spain	(Smith 1978)
Sweden	(Smith 1978)
Switzerland	(Smith 1978)
Ukraine (Ukrainian SSR)	(Smith 1978)

Potential Distribution within the US

Tremex fuscicornis is a Palearctic species, native to Asia and Europe. The species has successfully invaded New South Wales, Australia and the Área Metropolitana, Region V and Region VI of Chile. The insect was presumably introduced to Chile in infested wood packing materials from China. A coarse analysis of the worldwide distribution of this insect suggests it is most closely associated with temperate-broadleaf-and-mixed forests. This biome occurs in the northeastern US and accounts for 28% of the area within the contiguous US.

Survey

Few tools are available to aid surveys for *T. fuscicornis*. No sex pheromones or other chemical attractants have been identified. The insect is not known to be preferentially attracted by any colors. As a result, surveys must rely on visual inspections of the main stem of potential host trees for adult wasps, remnant ovipositors (rare), or emergence holes (reviewed in NAPPO 2006). Alternatively, bark can be removed to inspect for larvae or pupae, but this is not recommended if exit holes are not present. Intact trees or bolts of infested wood can be wrapped in aluminum screening to trap adults as they emerge from logs. Trees with drought stress or damaged by fire may be particularly predisposed to infestation (Parra S., unpublished).

Smith (2002) reported success in collecting siricids with a Malaise trap, which essentially is a large, open tent with a “roof” that slopes upwards into a collection vessel. Numerous versions of Malaise traps have been developed (Southwood 1966). Adult siricids are not commonly collected with such traps. They can be effective if adults are active and abundant (Smith and Schiff 2002).

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Fungus & Fungus-like Diseases

Gymnopus fusipes

Scientific Name

Gymnopus fusipes (Bull.) Gray, *Nat. arr. Brit. pl.* (London) 1: 604 (1821)

Fig. 1. Print of *Gymnopus fusipes* from 1818
[Image from The Royal Library, The National Library and Copenhagen University Library, http://www.pictures.dnlb.dk/FloraDanica/Hefte27/

Synonyms:

Agaricus crassipes Schaeff., *Icones*: tab. 87, tab. 88 (1762)
Agaricus fusiformis Bull., *Herbier de la France*: tab. 76 (1787)
Agaricus fusipes Bull., *Herbier de la France* 3: tab. 106 (1783) [1782-83]
Agaricus illicinus DC. in DC & Lam., *Fl. franç*. 5:48 (1815)
Agaricus lancipes Fr., *Hymenomyc. eur.* (Uppsala): 312 (1876)
Agaricus oedematopus Schaeff., *Fung. Bavar. Palat.* 4: 69 (1774)
Collybia contorta (Bull.) Raithelh., *Metrodiana* 8(2 -3): 52 (1979)
Collybia crassipes (Schaeff.) Ricken, *Die Blätterpilze*: 407 (1915)
Collybia fusipes (Bull.) Quél., *Mém. Soc. Émul. Montbéliard*, Sér. 2 5: 93 (1872)
Collybia fusipes var. *contorta* (Bull.) Gillet, *Hyménomycètes* (Alençon): 312 (1876)
Collybia fusipes var. *oedematopus* (Schaeff.) Gillet, *Hyménomycètes* (Alençon): 312 (1874)
Collybia illicina (DC.) Gillet, *Hyménomycètes* (Alençon): 313 (1876)
Collybia lancipes (Fr.) Gillet, *Hyménomycètes* (Alençon): 312 (1876)
Collybia oedematopoda (Schaeff.) Sacc., *Syll. fung.* (Abellini) 5: 206 (1887)
Rhodocollybia fusipes (Bull.) Romagn., *Bull. Soc. mycol. Fr.* 94: 78 (1978)

Common Names

root rot, *Collybia* root rot

Type of Pest

Fungus

Taxonomic Position

Kingdom: Fungi, **Phylum:** Basidiomycota, **Order:** Agaricales, **Family:** Tricholomataceae

Reason for inclusion in manual

Exotic Forest Pest Information System – classified as a very high risk pest with the potential to attack oaks

Pest Description

As described by Antonín and Noordeloos (1997), the genus *Gymnopus* has "basidiocarps fleshy; stipe fusoid, deeply longitudinally striate to sulcate, forming a distinct pseudorrhiza; spore print white to pale ochraceous; spores non-dextrinoid; cheilocystidia present; pileipellis a transition between cutis and trichoderm, made up of inflated, irregular, often coralloid elements, similar to the Dryophila-structure, often slightly gelatinized. Chemical reactions: no part of carpophores dextrinoid or cyanophilous."

The species *G. fusipes* has the following form: "pileus 30-90 mm broad, hemispherical, broadly conical to convex, expanding with age to broadly convex or plano-convex with low, broad umbo, with deflexed then straight or reflexed margin, hygrophanous, when moist translucently striate at margin only, dark red-brown or rusty brown ..., paler at centre and usually spotted with rusty or yellowish spots, pallescent upon drying to reddish yellow ..., glabrous, smooth to slightly rugulose, dull. Lamellae fairly distant, broadly adnate, sometimes somewhat emarginate, 4-8 mm broad, sometimes anastomosing, pale greyish brown, pale brown then dark brown or red-brown ..., often with small rusty spots, with entire, concolorous edge. Stipe 50-110 (-160 maximum observed) x 8-20 mm, fusiform, or more or less cylindrical above and fusoid in lower part, usually irregularly compressed and curved or flexuous, solid or narrowly fistulose, concolorous with lamellae at apex, downwards dark reddish brown ..., often with rust-coloured spots, strongly fibrillose-sulcate lengthwise, often twisted, glabrous or finely white-pruinose, dull or shining; at base originating from a root-like black sclerotium. Context whitish to sordid reddish. Smell indistinct, sometimes sweetish. Taste indistinct, fungoid. Spore print white, slightly turning yellowish on drying" (Antonín and Noordeloos 1997).

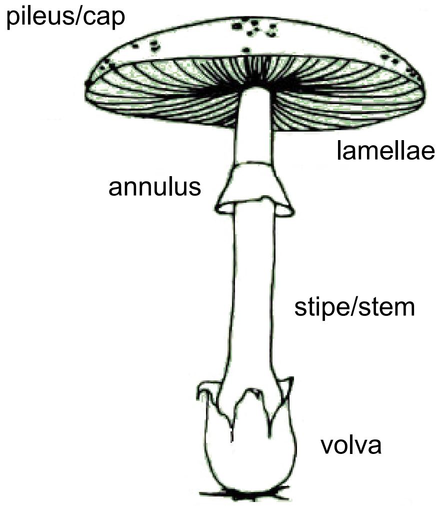

Fig. 2. Generalized drawing of fruiting body
[Image modified from David Largent, http://bcmushrooms.forrex.org/old-growth/Pages/framset.html. **Additional terms** defined at http://bcmushrooms.forrex.org/old-growth/Pages/framset.html]

Additional diagnostic features, including the morphology of spores, are described in detail by Antonín and Noordeloos (1997).

Gymnopus fusipes has two forms. Most commonly, it occurs as a saprophyte on dead plant material such as stumps or forest duff; alternatively, a pathogenic or “parasitic” form can be found near the base of the trunk on live, susceptible trees (Przybył 1994, Antonín and Noordeloos 1997). The two forms differ in their general appearance.

Saprophytic form: “Cap 20-40(-50) mm in diameter, convex, dark red-brown when moist, darker when dry. Stem 30-100 x 7-17 mm, light red-brown at apex, darker towards the base, with a swollen middle but tapering slightly towards a base, which can look like a long rooting base; the whole stem slightly grooved and twisted along its length. Basidia clavate, 25-35 x 5-7 µm with 2(-4) sterigmata. Basidiospores elliptic, exceptionally slightly pip shaped, 4.2-5.7 x 2.8-4.2 µm in size. Fruiting: end of August and beginning of September” (Przybył 1994).

Parasitic form: “Cap 30-60(-100) mm in diameter, slightly convex, light red-brown when moist, light brown when dry. Stem (50-) 70-120 (-130) mm, light brown at apex, darker towards the base; with swollen middle but tapering towards a rooting stipe: the whole stem visibly grooved and twisted along its length and fused at the base to several others. Basidia clavate, 25-40 x 5-8 µm with 4(-2) sterigmata. Basidiospores pip shaped, (4.2-)5.7 -7 x 2.8-5 µm in size. Fruiting: beginning of September (2 weeks later than the saprophyte)” (Przybył 1994).

A note on nomenclature. Nomenclature and taxonomy of *Collybia* and closely related genera has been highly confused. Halling (1983) and Antonín and Noordeloos (1997) provide historical accounts of revisions within and among genera. Recent molecular evidence supports the placement of *C. fusipes* within the genus *Gymnopus* (Antonín et al. 1997). The type species for *Collybia* is *C. tuberosa* (Bull:Fr.) Kummer. However, the placement of *C. fusipes* within *Gymnopus* is not universally accepted, and several authors continue to use “*Collybia fusipes*”.

Fig.3. Left, Fruiting body of *Gymnopus fusipes* viewed from above. Note umbo (raised surface) Right, *G. fusipes* viewed from the side.
[Images from Malcom Storey, http://www.bioimages.org.uk]

Biology and Ecology

Little is known about the biology, epidemiology and virulence of *G. fusipes* (Marçais and Delatour 1998).

Natural infection and colonization occurs when basidiospores germinate on the bark surface or by ectotrophic mycelial growth along the roots or bark or through the soil. Fruiting bodies or basidiomes are supported by mycelia, which grow from the infected host root tissue to the soil surface. *Gymnopus fusipes* is not known to form infection foci and does not spread by root grafts (Marçais and Delatour 1996, 1998, Marçais and Caël 2001, Camy et al. 2003b). Artificial inoculation studies have shown that *G. fusipes* is pathogenic on *Quercus robur* and *Q. rubra* saplings and apparently healthy, mature *Q. robur* trees (Marçais and Delatour 1996, 1998, Camy et al. 2003c). However, the pathogen is very slow growing and severe damage to roots may only become evident 30 years after the initial infection (Camy et al. 2003a). See 'Known Hosts' for a listing of the plants that can be infected by *G. fusipes* and 'Pest Importance' for a discussion of the impact the pathogen is having in Europe.

Marçais (2000) found that declining oaks with severe root damage were most common in areas with limited water holding capacity such as sandy soils layered over a deep, impenetrable gravel layer. A number of subsequent researchers also reported more severe infection by *G. fusipes* in acidic, sandy soils that did not regularly undergo seasonal flooding (Piou et al. 2002, Camy et al. 2003b, Camy et al. 2003c, Camy et al. 2003d). However, the cause-effect relationship between *G. fusipes* and oak decline is not clear. The pathogen may infect otherwise healthy oak roots, impede root function, and cause the tree to wilt (see 'Symptoms' for a complete list of the potential effects of the pathogen). Dry soils could accelerate the expression of symptoms. Alternatively, in the field, *G. fusipes* may be able to infect trees more effectively if trees are already stressed from dry soils. When *Q. robur* was introduced in Europe, it may have been planted in suboptimal sites, which may help to explain its apparent susceptibility to *G. fusipes* and oak decline in general (Camy et al. 2003b).

The pathogen itself seems to survive for the longest time on moist, well-drained soils. When pieces of hazel were colonized by *G. fusipes* and placed in sites of varying quality, the pathogen survived the longest in areas with coarse soils and low pH (<4.2). Inoculum survival was poor in heavily waterlogged soils (Camy et al. 2003c) due to induced hypoxic conditions (Camy et al. 2003c, Camy et al. 2003a, Camy et al. 2003d). In a greenhouse study involving waterlogged oak seedlings, *G. fusipes* did not survive beyond 21 days (Camy et al. 2003c, Camy et al. 2003d). In the field, *G. fusipes* is most abundant in coarse, dry to slightly hydromorphic soils (Camy et al. 2003b, Camy et al. 2003c).

Several other factors may influence the pathogenicity of *G. fusipes* including host susceptibility, tree health (age, condition), insect damage, environmental stresses (droughty or flooded soils) and abiotic site conditions (Guillaumin et al.

1983, Delatour and Guillaumin 1985, Guillaumin et al. 1985, reviewed in Przybył 1994, Marçais et al. 1999, 2000, Piou et al. 2002, Camy et al. 2003b, Camy et al. 2003c, Camy et al. 2003d).

The presence of *G. fusipes* is not necessarily associated with the decline of infected trees; host trees with heavily damaged root systems may have apparently healthy crowns (Marçais et al. 1999, 2000, Camy et al. 2003b). However, in one study from France, only 15% of trees with severely declining crowns were not infected by *G. fusipes* (Marçais et al. 1997). Although infected trees may not show signs of wilt, infected trees do experience a substantial reduction in growth rate (Marçais et al. 1997, Marçais and Caël 2001). Recent evidence suggests that *G. fusipes* does not preferentially infect stressed trees (Piou et al. 2002).

Pest Importance

Gymnopus fusipes is a soilborne root pathogen that has been isolated from declining European oak stands (Przybył 1994). The fungus typically occurs throughout its range as part of a complex of forest saprophytes and root rot pathogens such as *Ophiostoma quercus* and *Armillaria mellea* (Guillaumin et al. 1983, 1985 reviewed in Przybył (1994), Marçais and Caël 2001). *Gymnopus fusipes* has been documented as a saprophyte, an opportunistic pathogen that attacks stressed and weakened trees, a causal organism of root rot and even the causal organism in oak decline (Guillaumin et al. 1985, Marçais and Delatour 1996, 1998, Marçais et al. 1999, Camy et al. 2003b, Camy et al. 2003d). Research by Przybył (1994) notes *G. fusipes* as the causal organism for oak root disease, rather than oak decline. The relationship between these pathogens and their role in oak decline requires further study (Kowalski 1991, Przybył 1994, Marçais and Caël 2001).

The risks posed by *Gymnopus fusipes* for North American forests have been evaluated previously. The Exotic Forest Pest Information System (Cree 2004) considered the relative risks for this pest very high, but uncertain. *Gymnopus fusipes* may be a serious pathogen involved in oak decline in Europe, and similarly could pose a threat to oak forests in North America.

Symptoms

Gymnopus fusipes is a contributing factor to oak decline in Europe. Declining oaks typically have yellowing leaves, premature leaf drop, thin crown, branch dieback, epicormic shoots, bleeding cankers, tyloses formation, necrotic lesions in bark and phloem, discolored sapwood, and loosened bark (Kowalski 1991) Symptoms are similar to some other oak pathogens (see chapter on *Phytophthora quercina* in this volume). The cortical parenchyma in the root collar becomes necrotic turning yellowish-orange in color (Przybył 1994); the orange lesions on roots are considered uniquely symptomatic of the pathogen (Camy et al. 2003b). In more advanced stages of infection, abundant white mycelial fans occur within the inner bark and in the sapwood which appear wet and orange in color (Przybył 1994, Marçais et al. 1999).

The presence of fruiting bodies at the base of trees is a reliable indicator of *G. fusipes* infection. However, the presence of this fungus is not always associated with host decline (Marçais et al. 1999, 2000, Marçais and Caël 2001, Camy et al. 2003b). *Gymnopus fusipes* appears to grow and develop slowly, and symptoms of decline may not be apparent in infected trees for several months to several years (Marçais and Delatour 1996, Marçais et al. 1997, 2000, Marçais and Caël 2001).

Observations by Marçais (1999) and Marçais (2000) suggest that root damage begins in the central root system and progresses to the lateral roots where water and nutrient transport are affected. A decrease in sapwood width was found to be proportional to the level of infection. However, it is unclear whether host trees were in a state of decline and more susceptible to infection or if the infection caused the decline of healthy trees (Marçais et al. 2000). Marçais (2000) observed severe deterioration of tree crowns with severe infections and heavy root damage on some hosts, and insignificant differences in crown appearance and sapwood width, among other characteristics, for some neighboring damaged and undamaged oak trees in stands infected with *G. fusipes*.

In a survey of oak forests in Poland, Przybył (1994) noted symptoms of root rot on more than 200 infected oak trees (*Quercus robur* and *Q. petraea*) ranging from 70-120 years old. Before infection occurs, *G. fusipes* is present as a latent saprophyte on root surfaces (Guillaumin et al. 1985).

Known Hosts

Gymnopus fusipes occurs on deciduous tree hosts within the family Fagaceae, primarily on *Quercus* spp. (Delatour and Guillaumin 1985, Przybył 1994, Piou et al. 2002). Susceptibility to the pathogen apparently varies by host and may also be attributed to site factors (see 'Biology and Ecology'). In oak and chestnut seedling inoculation studies, *Quercus rubra*, a species native to North America, was more susceptible to infection by *G. fusipes* than *Q. petraea*, while susceptibility of *Q. robur* and *Castanea sativa* was intermediate (Marçais and Caël 2000). Similarly, *Quercus rubra* is noted as the most susceptible species in a survey of the reported hosts and distribution of *G. fusipes* in France (Piou et al. 2002).

Hosts	References
Acer sp. (maple)	reviewed in Piou et al. (2002)
Betula sp. (birch)	(Piou et al. 2002)
Carpinus betulus (hornbeam)	(Marçais and Caël 2000)
Carpinus carpinifolia (hornbeam)	(Piou et al. 2002)
Castanea crenata	(Piou et al. 2002)

Hosts	**References**
Castanea sativa (chestnut)	(Marçais and Caël 2000, Camy et al. 2003c)
Castanea sp. (chestnut)	(Kreisel 1961, USDA-ARS 2006)
Corylus avellana (hazelnut)	(Marçais and Caël 2000, Piou et al. 2002)
Corylus sp. (hazelnut)	(Kreisel 1961, Piou et al. 2002)
Fagus sp. (beech)	(Kreisel 1961, Piou et al. 2002, USDA-ARS 2006)
Fagus sylvatica (=*F. silvatica*) (European beech)	(Delatour and Guillaumin 1985, Piou et al. 2002, USDA-ARS 2006)
Pinus wallichiana[1] (Bhutan pine)	(USDA-ARS 2006)
Platanus sp. (sycamore)	(USDA-ARS 2006)
Quercus acutissima (sawtooth oak)	(Piou et al. 2002)
Quercus ×*bebbiana* [*alba* × *macrocarpa*] (white oak)	(Piou et al. 2002)
Quercus bicolor (swamp white oak)	(Piou et al. 2002)
Quercus cerris (European turkey oak)	(Piou et al. 2002)
Quercus coccinea (scarlet oak)	(Piou et al. 2002)
Quercus faginea (Portuguese oak)	(Piou et al. 2002)
Quercus ilex (holly oak)	(Piou et al. 2002)
Quercus imbricaria (laurel oak)	(Piou et al. 2002)
Quercus ×*heterophylla* (pro sp.) [*phellos* × *rubra*] (red oak)	(Piou et al. 2002)
Quercus incana (bluejack oak)	(USDA-ARS 2006)
Quercus myrsinifolia (Japanese evergreen oak)	(Piou et al. 2002)
Quercus palustris (pin oak)	(Piou et al. 2002)
Quercus petraea (sessile or durmast oak)	(Delatour and Guillaumin 1985, reviewed in CAB 2006)
Quercus phellos (willow oak)	(Piou et al. 2002)
Quercus pubescens (downy oak)	(Piou et al. 2002)
Quercus pyrenaica (Pyrenean oak)	(Piou et al. 2002)
Quercus robur (common, pedunculate oak)	(Delatour and Guillaumin 1985, reviewed in CAB 2006)
Quercus rubra (northern red oak)	(Delatour and Guillaumin 1985, reviewed in CAB 2006)
Quercus serrata (=*Q. glandulifera*)	(Piou et al. 2002)
Quercus suber (cork oak)	(Piou et al. 2002)
Quercus velutina (black oak)	(Piou et al. 2002)
Quercus × *vilmoriniana*	(Piou et al. 2002)

Hosts	References
Quercus sp. (oak)	(Kreisel 1961, USDA-ARS 2006)

1. Possible misidentification. *G. fusipes* is known to infect roots of deciduous trees.

Known Distribution

The pathogen has been widely reported from Europe. The presence of *Gymnopus fusipes* has not been confirmed in the US. There are a few historical records naming *G. fusipes*, but these can likely be attributed to taxonomic confusion (USDA-ARS 2006). In these records, *G. fusipes* was identified from the fruiting body stage on dead, dying or unspecified host material.

Location	References
Algeria (very rare)	(Piou et al. 2002)
Austria	(Antonín and Noordeloos 1997)
Belgium	(Antonín and Noordeloos 1997, USDA-ARS 2006)
Bulgaria	(Antonín and Noordeloos 1997, Piou et al. 2002)
Canary Islands	(Piou et al. 2002)
Czech Republic (Czechoslovakia, Slovakia)	(Antonín and Noordeloos 1997, USDA-ARS 2006)
Denmark	(USDA-ARS 2006)
England	(USDA-ARS 2006)
Finland (rare)	(Piou et al. 2002)
France	(Delatour and Guillaumin 1985, Piou et al. 2002, USDA-ARS 2006)
Germany	(Kreisel 1961, Piou et al. 2002)
Great Britain (rare)	(Piou et al. 2002)
Greece	(USDA-ARS 2006)
India	(USDA-ARS 2006)
Ireland	(Muskett and Malone 1980, USDA-ARS 2006)
Italy	(Antonín and Noordeloos 1997, Piou et al. 2002, USDA-ARS 2006)
Latvia	(USDA-ARS 2006)
Luxembourg	(Piou et al. 2002)
Mexico	(Guzman et al. 1992)
Morocco (very rare)	(Piou et al. 2002)
Netherlands	(Antonín and Noordeloos 1997, Piou et al. 2002)
Pakistan	(USDA-ARS 2006)
Poland	(Przybył 1994)

Location	References
Romania	(Antonín and Noordeloos 1997, reviewed in Piou et al. 2002, USDA-ARS 2006)
Spain	(Piou et al. 2002)
Sweden (rare)	(Antonín and Noordeloos 1997, Piou et al. 2002)
Ukraine	(Antonín and Noordeloos 1997)
United Kingdom	(Antonín and Noordeloos 1997)
United States[1]	(USDA-ARS 2006)

[1] Possible misidentification or taxonomic confusion. *G. fusipes* is not known to occur in the US.

Potential Distribution within the US

A coarse climatic analysis based on the reported geographic distribution of *G. fusipes* suggests the pathogen may be associated with temperate-broadleaf-and-mixed forests and tropical-and-subtropical-dry-broadleaf forests. These two biomes account for approximately 28% of the area within the contiguous US, primarily east of the Mississippi River. Algeria and Morocco, where *G. fusipes* is very rare, do not follow this pattern. In these countries the pathogen may be associated with temperate-coniferous forests, desert-and-xeric shrublands, or Mediterranean scrub.

Survey

Few tools are available to simplify surveys for *G. fusipes*. Although it may be tempting to focus surveys exhibiting symptoms of oak decline, such surveys are unlikely to locate the pathogen while it is still spatially confined. Several months to years may elapse between infection and the expression of symptoms (see ‘Symptoms’).

A version of visual survey will be needed to locate the pathogen in the field. The base of trees can be inspected for the mushrooms (basidiomes) of *G. fusipes*. Basidiomes at the base of a tree provide an indication of the degree of infection (Piou et al. 2002). Alternatively, in an effort to detect the pathogen early, roots of trees on sandy soils that are not waterlogged should be inspected. Soil should be removed from the root collar and major roots to a depth of 20-30 cm [8-12 in] and 80-100 cm [32-39 in] from the base of a tree (Marçais and Delatour 1998, Marçais et al. 2000). Samples of potentially infected roots should be collected from up to 12 locations (Marçais and Delatour 1998).

The number of trees to be inspected will depend on the expected frequency of infections. In heavily infected stands, 40% of the trees may be affected (Marçais et al. 1997).

To confirm the presence of the pathogen, orangish-yellow sapwood or white mycelium should be placed on semi-selective MAT medium (15 g agar, 10 g malt, 250 mg thiabendazole, 100 mg penicillin, 100 mg streptomycin, and 1 L distilled water (Marçais and Delatour 1998). A well trained mycologist will be needed to confirm identification (Cree 2004).

Halling (2004) provides a useful key to four genera of Agaricales found in the northeastern US:
"1. Spore deposit pinkish cream to pinkish buff when fresh; some spores cyanophilous and dextrinoid; basidiocarps typically putrescent and fleshy... *Rhodocollybia*
1. Spore deposit white to ivory yellow or buff, rarely pink; spores neither cyanophilous nor dextrinoid; basidiocarps membranous to marcescent..............2

> 2. Basidiomata white to gray, typically arising from a sclerotium or blackened fungous remains, rarely in humus, with filiform stipe and small pileus (<20 mm); spores typically less than 6 x 3 µm................................3
>
> > 3. Stipe producing narrow side branchlets with conidia; sclerotia black to dark gray .... *Dendrocollybia*
> >
> > 3. Stipe lacking side branchlets; sclerotia dark yellow, orange to reddish brown or absent.. *Collybia*
>
> 2. Basidiomata neither arising from a sclerotium nor blackened fungous remains, if in humus, stipe never filiform and with pileus >20 mm; spores usually >6 µm.. *Gymnopus*"

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Ophiostoma quercus
a hardwood-inhabiting species of the *O. piceae* complex

Scientific Name
Ophiostoma quercus (Georgévitch) Nannfeldt 1926

Synonyms:
Ophiostoma querci (Georgévitch) Nannf. 1934
Ceratostomella querci Georgévitch 1926
Ceratocystis quera (Georgévitch) Moreau 1952
Ophiostoma fagi (Loos) Nannf. 1934
Ceratostomella fagi Loos 1932
Ceratocystis fagi (Loos) Moreau 1952
? *Ophiostoma roboris* Georgescu & Teodoru 1948
Ceratocystis roboris (Georgescu & Teodoru) Potlajchuk 1985
? *Ophiostoma valachicum* Georgescu & Teodoru 1948
Ceratocystis valachicum (Georgescu & Teodoru) Potlajchuk, 1985
? *Ophiostoma kubanicum* Sherbin-Parfenenko 1953
Ceratocystis kubanicum (Sherbin-Parfenenko) Potlajchuk 1985
? *Graphium kubanicum* Potlajchuk & Schekunova 1985

Anamorph: *? Graphium roboris* Georgescu & Teodoru 1948
Synanamorph: *Sporothrix* sp.

Common Names
Blue stain or sapstain fungus (both common names also are used for other closely related fungi)

Type of Pest
Fungal pathogen and saprophyte

Taxonomic Position
Kingdom: Fungi, **Phylum:** Ascomycota, **Order:** Ophiostomatales, **Family:** Ophiostomataceae

Reason for inclusion in manual
CAPS Priority Pest (FY 2007) - listed as *O. piceae*

Pest Description
Considerable taxonomic confusion surrounds the ophiostomatoid fungi due to similarities in morphology, host and vector associations (Zipfel et al. 2006); *O. piceae* in particular has been considered a species complex (Harrington et al. 2001). *Ophiostoma quercus* was only recently identified as a distinct species. '*Ophiostoma piceae'* from hardwoods (OPH) was found to be reproductively isolated from '*O. piceae*' isolated from conifers (OPC), but this distinction was

(and remains) imperfect (Brasier 1993, Brasier and Kirk 1993, Harrington et al. 2001). The OPH breeding group was later described as a distinct species, *O. quercus*, by Morelet (1992). Some authors have used the similar name *O. querci*, but this usage is incorrect (De Beer et al. 2003b).

Older literature treats *O. piceae* and *O. quercus* as a single species. Because *O. quercus* primarily affects hardwood species and *O. piceae* primarily affects conifers, we infer that older reports of *O. piceae* on hardwoods are actually describing *O. quercus*. The opposite case is not as likely because as there are several confirmed cases of *O. quercus* being isolated from conifers (Harrington et al. 2001).

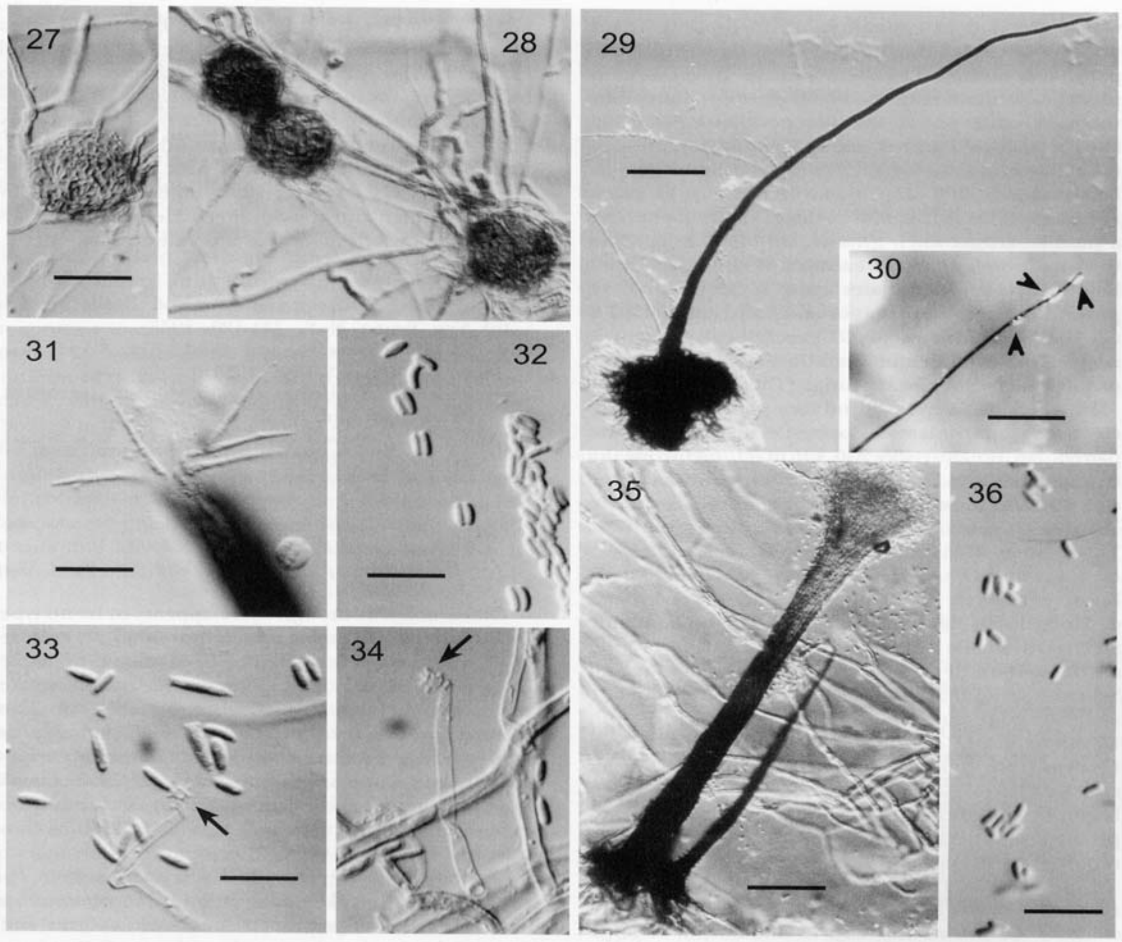

Fig. 1. Photomicrographs of *Ophiostoma quercus*. "27, 28. Protoperithecia. 29. Perithecium. 30. Perithecial neck with drops of ascospores (arrowheads). 31. Ostiolar hyphae. 32. Ascospores. 33, 34. Conidiophores and conidia of the *Sporothrix* synanamorph. Arrows in 33 and 34 indicate apex of conidiogenous cells, showing prominent denticles. 35. Synnema. 36. Conidia from synnema. 27, 28, 33-36 from strain C969; 29-32 from a pairing of 969 and C970. Scale bars: 27, 28 = 25 μm; 29 = 100 μm; 30 = 220 μm; 31-34, 36 = 10 μm; 35 = 50 μm."

[Image and portions of caption reproduced from Harrington et al.(2001)]

Ophiostoma quercus is closely related to other species within the genera *Ophiostoma, Ceratocystis, Ceratocystiopsis*, *Graphium, Leptographium,* and *Pesotum*. The genus designation depends on whether a sexual form (teleomorph) or asexual form (anamorph) of the fungus is being described.

Ophiostoma quercus is most easily distinguished from other ophiostomatoid fungi by characteristics of its growth on malt extract agar: light-brown color of the protoperithecia, mycelium with concentric rings (most isolates), a nutty smell, and 5-10 mm of growth after 7 days at 89.6°F [32°C] (Harrington et al. 2001).

Ophiostoma quercus "is difficult to distinguish morphologically from *O. piceae*, though ITS sequencing shows it to be only [*sic*] distantly related. It is common to find small, glistening drops of ascospores along the neck of *O.* [*quercus*] perithecia (Fig. 1-30), which are due to exudations of ascospores as the perithecial neck elongates. We have not seen this in perithecia of the other species in the *O. piceae* complex, but we have seen such ascospore droplets along the neck of *O. pluriannulatum* perithecia. Most isolates of *O.* [*quercus*] and *O. piceae* form protoperithecia on [malt extract agar] MEA (Fig.1-27, 28), but those of *O.* [*quercus*] are a light, golden-brown color, while those of *O. piceae* are black. Most isolates of *O.* [*quercus*] have a nut-like aroma when growing on MEA, in contrast to an indistinct aroma in cultures of *O. piceae*. Also, *O.* [*quercus*] isolates grow at 32°C [90°F], while *O. piceae* isolates do not (Brasier and Stephens 1993). Many isolates of *O.* [*quercus*] form concentric rings of aerial mycelium on MEA, similar to isolates of the Dutch elm disease fungi" (Harrington et al. 2001).

"In pairings, *O.* [*quercus*] produces perithecia and ascospores when isolates of opposite mating types are paired." (Harrington et al. 2001).

When grown on 3% malt agar, colonies of *O quercus* were 55-67 mm in diameter after 10 days at 25°C [77°F] in the dark (Przybył and Morelet 1993). Little aerial mycelia was produced. Mycelia was "floccose usually growing in sectors [and] whitish grey" (Przybył and Morelet 1993). "Synnemata were produced abundantly all over the colonies or in concentric zones" (Przybył and Morelet 1993). Branched or unbranched synnemata were (130-) 350-500 (-600) μm long; conidiogenous cells within synnemata were 8.3-16.6 μm long (Przybył and Morelet 1993). Synnematal conidia were "oblong to globose", appearing similar to yeast-like cells, and readily germinated within 3 days on SNA medium at 25°C in the dark (Przybył and Morelet 1993). Bases of ascocarps produced on sapwood were between 90-160 (-190) μm; neck hyphae were 1,100-1,900 (-2,500) μm long; ostiolar hyphae were (11.6-) 16.6-36.5 μm long (Przybył and Morelet 1993). Ascospores were (3.5-) 2.5x1.5 μm and were "allantoid in side view" (Przybył and Morelet 1993).

Biology and Ecology

Mycelial growth depends on temperature and relative humidity (RH). Optimal growth of mycelia of *O. quercus* occurs between 22.5-27.5°C [72.5-81.5°F]; the upper limit for the growth of mycelia is approximately 35°C [95°F] (Brasier and Stephens 1993). Humidity requirements for the growth of *O. quercus* have not been reported, but Payne (2000) found a minimum 93-94% RH was needed for the growth of *O. piceae* on laboratory growth medium and *Picea* sp. sapwood.

O. quercus reproduces sexually by ascospores produced in perithecia and asexually by conidia produced in synnemata. Sexual reproduction requires two mating types, which have been designated A and B (Brasier and Kirk 1993, Brasier and Stephens 1993).

Ophiostomatoid fungi are dispersed by bark beetles (Coleoptera: Scolytidae), other phloem-feeding wood borers, and rain splash (Gibbs 1993-Biology; Gagné et al. 2001; Jankowiak 2005; Campbell 1960; Graham 1967). Scolytids are known to have mutualistic relationships with ophiostomatoid fungi, whereby the fungus is dispersed by the insect when the insects bore into new hosts. The fungus is presumably used by the insect either directly as a food source or as a digestive mechanism that breaks down host plant tissues (Baker 1963, Graham 1967, Nevill and Alexander 1992, Harrington 1993, Paine et al. 1997, Yamaoka et al. 1997, Yamaoka et al. 1998, Jacobs et al. 2002, Kirisits et al. 2002, Jacobs et al. 2003, Jankowiak 2005). Fragments of hyphae/perithecia may be transported in mycangia (specialized integumentary pores in the pronotum) and on the outer body surface of the insect vector. *Ophiostoma* spp occur in the tunnels and galleries created by the insect and will grow into phloem, sapwood and heartwood (ray and parenchyma cells) to obtain nutrients. In this process, the fungus stains the wood (Wilson 1959, Baker 1963, Graham 1967, Harrington 1993, Gharibian et al. 1996, Abraham et al. 1998, Bruce et al. 2003). In West Virginia, *O. quercus* has been isolated from larval cradles, main galleries and bodies of the timber beetle *Corthylus columbianus* in *Quercus alba* (white oak); this beetle is endemic to North America (Wilson 1959).

See ‘Known Hosts’ for a list of plants that can be infected by *O. quercus* and ‘Pest Importance’ for a discussion of the economic impact of this pathogen.

Pest Importance

Ophiostoma quercus is a pathogen when it infects living trees and a saprophyte when it grows on down or cut woody material. Most ophiostomatoid fungi are weak pathogens (Kim et al. 2005), but the virulence of *O. quercus* has been debated. Harrington (1993) and Jankowiak (2005) suggest species of *Ophiostoma* are weakly pathogenic, while Jacobs (2002) describes members of the genus as “serious pathogens.” Nevill and Alexander (1992) contend that the *O. piceae*-*O. quercus* complex is of intermediate pathogenicity. In an inoculation study by Przybył (1990a), *Quercus robur* seedlings died within 8 months following inoculation with three strains of *O. quercus*.

Ophiostoma spp. have been associated with both abiotic and biotic tree diseases, however the extent to which *Ophiostoma* can be attributed to tree death is not well understood. Because little is known about beetle-fungus associations, the extent of tree mortality that can be attributed solely to insect damage is also unclear (Harrington 1993). Host-resistance mechanisms triggered by a combination of abiotic and biotic factors should also be considered (Gibbs 1993).

Ophiostoma quercus has been isolated from dying oak trees in Europe (Harrington 1993). In Hungary and Slovakia, increased temperature and decreased soil moisture facilitate the transmission of *O. quercus*-related oak vascular mycosis by the bark beetle *Scolytus intricatus* (EPPO 1990). In Italy, *O. quercus* has been isolated from tissues of *Quercus cerris* and *Q. pubescens* affected by oak decline (EPPO 1990). *O. quercus* was found in necrotic lesions on the trunks of *Quercus robur* in Poland (Przybył 1990a). Although *Ophiostoma* species, including *O. quercus*, are present in declining oak stands in Europe, they probably do not play a major role in initiating decline within healthy oak stands (EPPO 1990).

Species of *Ophiostoma* and *Ceratocystis* are among the first fungi to appear on freshly cut wood, though *O. piceae* can be isolated readily from wood up to eighteen months after felling (Seifert 1993). Uzunovic et al. (1999) suggest that *O. piceae* appears to be spread by sawing through logs in mills where this fungus is present. It is likely that these properties also apply to *O. quercus*.

Sapstaining fungi cause significant worldwide economic losses in the forest products industry (Gagné et al. 2001, Kim et al. 2005). Stained wood is unsightly and may be rejected by the timber buyer and the end-user of the wood (Seifert 1993, Uzunovic et al. 1999, Payne et al. 2000). Many other fungi are known to grow alongside sapstain fungi on a single piece of wood, which may lead to discoloration as well as decay and loss of strength (Seifert 1993, Payne et al. 2000). Most sapstaining fungi metabolize non-structural components of the sapwood, such as sugars, proteins, and triglycerides while structural components like lignin and cellulose are left untouched (Gharibian et al. 1996, Abraham et al. 1998, Bruce et al. 2003). As a result, the damage to affected lumber is primarily cosmetic (Seifert 1993, Bruce et al. 2003), but these impacts vary with the tree species. Seifert (1993) summarizes some changes in wood properties caused by "*O. piceae*", but some of these observations, especially for *Betula* sp., likely refer to *O. quercus*. The fungus caused reductions in weight, specific gravity, compression parallel to grain, modulus of rupture, total work to bend, total work to maximum load, and toughness. The property most strongly affected was toughness, which was decreased by up to 25%. Other losses were considered insignificant.

Risks associated with *O. quercus* have not been evaluated formally, but Hildebrand (2005) discusses potential risks associated with *O. piceae*, distinct from *O. quercus*. Like *O. quercus*, *O. piceae* is already widely established in North America, so any new risks come from unknown variants of the fungus. Risks from these new variants were considered high but very uncertain (Hildebrand 2005), primarily because both the potential for establishment and spread were considered high. However, this assessment is highly speculative and should be treated with caution, as molecular and biological differences among most established strains of *O. quercus* have yet to be determined.

Symptoms

Sapstain is a blue, grey, or black discoloration of cut wood, caused mainly by several species of Ascomycete fungi in the genera *Ophiostoma* and *Ceratocystis* (Luck et al. 1990, Uzunovic et al. 1999). Melanized hyphae penetrate into the ray parenchyma, resin ducts, and cell lumens of affected wood, causing it to appear discolored (Seifert 1993, Uzunovic et al. 1999, Payne et al. 2000, Bruce et al. 2003). However, no actual staining of the cell walls occurs (Seifert 1993). Seifert (1993) explains the discoloration as an “optical effect” of the fungal melanin granules in the cells. In trees and freshly cut logs, growth of fungal mycelia along the medullary rays causes a triangular or wedge-shaped stain in cross-section (Gibbs 1993). There are no known external symptoms directly attributed to *O. quercus*.

Known Hosts

Hosts	References
Acer sp. (maple)[1]	(De Beer et al. 2003a)
Betula sp. (birch)[1]	(Seifert 1993)
Betula pubescens (downy birch)[1]	(Upadhyay 1981)
Betula platyphylla (Asian white birch)	(De Beer et al. 2003a, USDA-ARS 2006)
Cupressocyparis macrocarpa	(De Beer et al. 2003a)
Eucalyptus sp. (eucalyptus)	(Harrington et al. 2001, De Beer et al. 2003a, USDA-ARS 2006)
Eucalyptus grandis	(De Beer et al. 2003a, USDA-ARS 2006)
Fagus sp. (beech)	(Pipe et al. 1995, Harrington et al. 2001)
Fagus japonica (Japanese beech)[1]	(CAB 2006)
Fagus sp.	(USDA-ARS 2006)
Fagus sylvatica (European beech)	(De Beer et al. 2003a)
Juglans cinerea	(USDA-ARS 2006)
Kalopanax sp. (castor aralia)[1]	(De Beer et al. 2003a)
Laurelia philippiana (tepa)[1]	(De Beer et al. 2003a)
Laurelia sempervirens (tihue)[1]	(De Beer et al. 2003a)
Macaranga capensis (river macaranga)[1]	(Van Wyk and Wingfield 1992)

Hosts	**References**
Magnolia sp. (magnolia)[1]	(De Beer et al. 2003a)
Nothofagus sp. (southern beech)	(Butin and Aquilar 1984, Paclt 1985)
Nothofagus dombeyi (coigue)[1]	(De Beer et al. 2003a)
Nothofagus fusca (red beech)[1]	(De Beer et al. 2003a, USDA-ARS 2006)
Nothofagus pumilio (lenga beech) [1]	(De Beer et al. 2003a)
Olinia sp.	(De Beer et al. 2003a)
Olinia emarginata	(USDA-ARS 2006)
Olinia radiata	(USDA-ARS 2006)
Olinia ventosa	(De Beer et al. 2003a, USDA-ARS 2006)
Pinus sp. (pine) (including chips)	(Brasier and Kirk 1993, Pipe et al. 1995, Harrington et al. 2001, De Beer et al. 2003a, USDA-ARS 2006)
Prunus sp. (plum)[1]	(De Beer et al. 2003a)
Pseudotsuga sp. (Douglas-fir)	(USDA-ARS 2006)
Pyrus communis (pear)[1]	(Hausner and Reid 2003)
Quercus sp. (oak)	(Brasier 1990, Brasier and Kirk 1993, Pipe et al. 1995, Harrington et al. 2001, De Beer et al. 2003a, USDA-ARS 2006)
Quercus alba (white oak)[1]	(Wilson 1959)
Quercus longipes (long-stalk oak)	(Pipe et al. 1995, USDA-ARS 2006)
Quercus petraea (durmast oak)[1]	(EPPO 1990, CAB 2006)
Quercus robur (common oak)	(De Beer et al. 2003a, USDA-ARS 2006)
Tsuga sp. (hemlock)	(Pipe et al. 1995, Kim and Breuil 2001, De Beer et al. 2003a, USDA-ARS 2006)
Ulmus carpinifolia (English elm)	(Pipe et al. 1995, USDA-ARS 2006)

1. Original report refers to "*Ophiostoma piceae*" but may actually describe *O. quercus*. *Ophiostoma piceae* generally does not infect hardwoods, except *Ulmus* sp, *Acer* sp., and *Populus* sp. (Brasier and Kirk 1993).

Known Distribution

Ophiostoma quercus may be native to Europe and introduced into North America and the Southern Hemisphere (reviewed in De Beer et al. 2003a). However, recent evidence casts doubt on this idea (De Beer et al. 2003a). Nevertheless, the species is now common and widespread, especially in the Northern Hemisphere, including North America. The table below summarizes countries where *O. quercus* specifically has been detected or infection of hardwoods (other than *Ulmus* sp., *Acer* sp., and *Populus* sp.) has been reported.

Location	**References**
Austria	(EPPO 1990, CABI/EPPO 2002, De Beer et al. 2003a, Hildebrand 2005,

Location	References
	CAB 2006)
Australia	(De Beer et al. 2003a)
Azerbaijan	(Brasier and Kirk 1993, Brasier and Stephens 1993, Pipe et al. 1995, Hildebrand 2005, CAB 2006)
Belgium	(Hildebrand 2005, CAB 2006)
Brazil	(De Beer et al. 2003a)
Bulgaria	(CABI/EPPO 2002, Hildebrand 2005, CAB 2006)
Canada	(Brasier and Kirk 1993, Pipe et al. 1995, Abraham et al. 1998, Uzunovic et al. 1999, Gagné et al. 2001, Kim and Breuil 2001, CABI/EPPO 2002, De Beer et al. 2003a, Hausner and Reid 2003, Jacobs et al. 2003, Hildebrand 2005, CAB 2006, USDA-ARS 2006)
Chile	(Butin and Aquilar 1984, CABI/EPPO 2002, De Beer et al. 2003a, Hildebrand 2005, CAB 2006, USDA-ARS 2006)
Czech Republic	(EPPO 1990, Przybył and Morelet 1993, CABI/EPPO 2002, Hildebrand 2005, CAB 2006)
France	(Brasier and Kirk 1993, Brasier and Stephens 1993, Przybył and Morelet 1993, Pipe et al. 1995, CABI/EPPO 2002, De Beer et al. 2003a, Hildebrand 2005, CAB 2006)
Germany	(EPPO 1990, CABI/EPPO 2002, De Beer et al. 2003a, Hausner and Reid 2003, Hildebrand 2005, CAB 2006, USDA-ARS 2006)
Hungary	(EPPO 1990, Brasier and Kirk 1993, Brasier and Stephens 1993, CABI/EPPO 2002, CAB 2006)
Italy	(EPPO 1990, CABI/EPPO 2002, Hausner and Reid 2003, Hildebrand 2005, CAB 2006)
Japan	(Yamaoka et al. 1997, Kim and Breuil 2001, CABI/EPPO 2002, De Beer et al. 2003a, Hildebrand 2005, Kim et al. 2005, CAB 2006)
Korea	(Kim et al. 2005)
Netherlands	(EPPO 1990)

Location	**References**
New Zealand	(CABI/EPPO 2002, De Beer et al. 2003a, Hildebrand 2005, Kim et al. 2005, CAB 2006)
Poland	(Przybył 1990b, Brasier and Kirk 1993, Brasier and Stephens 1993, Przybył and Morelet 1993, Pipe et al. 1995, Kim and Breuil 2001, CABI/EPPO 2002, De Beer et al. 2003a, Hildebrand 2005, Jankowiak 2005, CAB 2006)
Romania	(Brasier and Kirk 1993, Brasier and Stephens 1993, CABI/EPPO 2002, Hildebrand 2005, CAB 2006)
Russia	(CABI/EPPO 2002, Hildebrand 2005, CAB 2006)
Slovakia	(EPPO 1990, CABI/EPPO 2002, Hildebrand 2005, CAB 2006)
South Africa	(CABI/EPPO 2002, De Beer et al. 2003a, Hildebrand 2005, CAB 2006)
Sweden	(Kim and Breuil 2001, Hildebrand 2005)
Tajikistan	(Brasier and Kirk 1993, Brasier and Stephens 1993, Pipe et al. 1995)
United Kingdom	(Brasier and Kirk 1993, Brasier and Stephens 1993, Pipe et al. 1995, Pipe et al. 2000, Kim and Breuil 2001, CABI/EPPO 2002, De Beer et al. 2003a, Hildebrand 2005, CAB 2006)
United States	(Davidson 1953, Wilson 1959, Campbell 1960, CABI/EPPO 2002, De Beer et al. 2003a, Hausner and Reid 2003, CAB 2006)
Uruguay	(Harrington et al. 2001, De Beer et al. 2003a)

Potential Distribution within the US

Ophiostoma quercus is widely established in North America. This distribution is not surprising given the habitat types with which the pathogen appears to be associated outside the US. The pathogen is associated with temperate-broadleaf-and-mixed forest through most of Europe and eastern Asia. However, this biome does not occur in Uruguay, Brazil, Tajikistan, or South Africa. In these four countries, the fungus is likely associated with desert-and-xeric shrublands and temperate grasslands, savannas, and shrublands. Collectively, these three biomes account for 80% of the area within the contiguous US.

Survey

Surveys for this fungus are likely to be very difficult and will require well trained personnel to process samples and interpret results.

Many species of *Ophiostoma* and *Ceratocystis* occur between bark and sapwood near areas that are freshly wounded, in galleries created by bark beetles, or in diseased roots (Seifert et al. 1993). Inspection of trees with a hand lens often will reveal "slimy spore masses at the tip of black stalks" (Seifert et al. 1993). Wood samples should be processed soon after collection. Samples should be held at room temperature in Petri dishes with moistened filter paper (Seifert et al. 1993, De Beer et al. 2003a). Spores should be collected from perithecia or conidiophores as these structures emerge and transferred to malt extract agar. The addition of the antibiotic cyclohexamide to media will aid isolation of the fungus. Standard microbiological techniques should be used to purify cultures (De Beer et al. 2003a).

Isolates may also be identified by vegetative incompatibility (mating) reaction patterns, variation of colony types; and electrophoretic patterns of soluble proteins (Brasier 1993, Brasier and Kirk 1993). For mating compatibility studies, a single ascospore or conidium from each of the two colonies to be tested should be transferred to water agar (30 g agar/L distilled water) with two small pieces of oak wood (each 5 x 5 x 25 mm) (De Beer et al. 2003a). Dishes should be held at room temperature until perithecia develop. 'Tester strains' can be identified from the relative abundance of perithecia produced in these assays. Perithecia will only be produced when both A and B types of the same species are present.

It is also possible to distinguish species using molecular techniques. Harrington et al. (2001) amplified ribosomal DNA (ITS) with polymerase chain reaction (PCR) and cut the PCR product with the restriction enzyme HaeII to produce a unique banding pattern on an agarose gel. More detailed protocols are provided by Brasier (1993), De Beer (2003a), and Pipe (1995). Kim et al. (1999) provide a useful protocol to quickly detect and identify *O. quercus* on wood using PCR.

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Phytophthora quercina

Scientific Name
Phytophthora quercina Jung, Cooke, Blaschke, Duncan, and Oswald

Synonyms:
none known

Common Names
Phytophthora root rot (a common name applicable to many *Phytophthora* spp)

Type of Pest
Fungus-like

Taxonomic Position
Kingdom: Chromista, **Phylum:** Oomycota, **Order:** Pythiales,
Family: Pythiaceae

Reason for inclusion in manual
CAPS Priority Pest (FY 2007)

Pest Description
Distinctive morphological features of *P. quercina* are not visible with the unaided eye. Even when examined under a microscope, the size, shape, and appearance of hyphae, sporangiophores, sporangia, oogonia, oospores, antheridia, and chlamydospores vary considerably for *P. quercina* (Jung et al. 1999).

Sporangia – Sporangia bear a lump or swelling and are egg shaped (ovoid), round (globose), nearly round (sub-globose), pear-shaped, with broad end proximal (obpyriform), flask like (ampulliform), or “banana- or peanut-like” (Fig. 1) (Jung et al. 1999). Dimensions: 19-112 µm long, 14-47 µm wide.

Fig. 1. Sporangia of *Phytophthora quercina*
[Image from T. Jung, 2000.
http://www.forst.uni-muenchen.de/EXT/LST/BOTAN/LEHRE/PATHO/QUERCUS/oakdec.htm]

Chlamydospores – Chlamydospores may not be observed because they are not produced consistently. When grown on malt extract agar, chlamydospores are spherical, 17-35 µm diameter (Jung et al. 1999).
Oospores – Oospores are globose, 18- 38 µm diameter (Jung et al. 1999).
Oogonia – Oogonia are irregularly shaped, spherical to ovoid, 19-45 µm diameter and up to 52 µm long (on malt extract agar) (Jung et al. 1999). Isolates from France had a slightly different appearance, with oogonia sometimes enveloped in a sheath (Hansen and Delatour 1999).

Phytophthora quercina is in Group I of the Waterhouse (1970) key to *Phytophthora* and is distinguishable from species in Group III by the thickness and shape of the papilla (Jung et al. 1999), a nipplelike projection at the apical end of the sporangium. Group I species have conspicuous papillae that are ~3.4 µm thick, while papillae of Group III species are less conspicuous, only ~2.1 µm thick. Unlike Group IV species, *P. quercina* has paragynous antheridia and sporangia that are easily dislodged from sporangiophores (Jung et al. 1999). Group IV species also have lower optimum and maximum temperatures for growth compared to Group I species. The sporangia of *P. quercina* look a bit like *P. nicotianae* from Group II, however *P. nicotianae* has amphigynous antheridia and other characters which are distinguishable from *P. quercina* (Jung et al. 1999).

Biology and Ecology

The biology of *P. quercina* remains poorly described (Cree 2005), partly because the species is still relatively new to science (Jung et al. 1999). Mycelia will grow at a wide range of temperatures (between 5-27.5°C [41-81.5°F]); however, optimal growth occurs between 22.5-25°C [72.5-77°F] (Jung et al. 1999, Barzanti et al. 2001).

A single isolate can produce numerous oospores, which can survive in a state of dormancy for several years (Jung et al. 1999). *Phytophthora quercina* has reportedly survived for 2 years at 18°C [64°F] in dry oak forest soil (Cooke et al. 2005). The pathogen is homothallic, so oospores can be produced if only one mating type is present.

Though this pathogen can grow under a wide variety of conditions, drier sites with nutrient-rich clayey and loamy soils and soil pH ranging from 3.5-7.0 appear to be most favorable (Jung et al. 2000, Hartmann and Blank 2002, Balci and Halmschlager 2003a, Jonsson et al. 2005). Higher concentrations of calcium and magnesium may favor the development of the pathogen and indirectly facilitate the infection process (Balci and Halmschlager 2003a).

See ‘Known Hosts’ for a list of plants that can be infected by *P. quercina* and ‘Pest Importance’ for a discussion of the impact the pathogen is having in Europe.

Pest Importance

Phytophthora quercina has been isolated from declining European oak stands (Hansen and Delatour 1999, Schubert et al. 1999, Balci and Halmschlager 2002, Hartmann and Blank 2002, Vettraino et al. 2002). In Germany, annual oak mortality [attributed in part to oak decline] is estimated at 2-5 mature oaks/ha [ca. 1-2 oaks/acre] (Heiser et al. 1999). *Phytophthora quercina* is predominantly a soilborne root pathogen (Jung et al. 1999, Balci and Halmschlager 2003a, Jonsson et al. 2003). In greenhouse inoculation studies, the pathogen was able to kill 35-50% of the fine roots of year-old oak (Q*uercus robur*) seedlings in <4 months (Jung et al. 1996). Minor lesions (10-23 mm after 3 months) developed on *Q. robur* when stems were injected with the pathogen (Jung et al. 1996). However, another isolate of the pathogen was unable to infect the stem or collar of *Q. petraea* seedlings in a separate greenhouse inoculation test (Balci and Halmschlager 2003a).

In previous pest risk assessments, the pathogen was considered moderately likely to invade the US and cause economic harm (Cree 2005); the pathogen was considered relatively unlikely to cause environmental harm that could be distinguished from other causes of oak decline in the US. Even in Europe, the relationship between oak decline and *P. quercina* is not absolute. Although the pathogen is frequently isolated from stands with oak decline (Hansen and Delatour 1999, Schubert et al. 1999, Jung et al. 2000, Balci and Halmschlager 2002, Hartmann and Blank 2002, Vettraino et al. 2002), it is also isolated from apparently healthy stands (Hansen and Delatour 1999, Jung et al. 2000, Balci and Halmschlager 2003b). Pathogens other than *P. quercina* may be isolated from stands with oak decline (Jung et al. 1996, Vettraino et al. 2002, Balci and Halmschlager 2003b, Jonsson et al. 2003). The pathogenicity of *P. quercina* may depend on other predisposing factors, such as drought, flooding, defoliation, and tree species composition (Hansen and Delatour 1999, Schubert et al. 1999, Balci and Halmschlager 2002, Hartmann and Blank 2002, Vettraino et al. 2002). Alternatively, *P. quercina* may be an inciting factor that leaves a tree vulnerable to other types of environmental stress that alone would be inadequate to kill a tree (Vettraino et al. 2002).

Symptoms

Symptoms of infection by *P. quercina* are similar to those caused by other pathogens associated with oak decline: leaf clusters; twig abscission; epicormic shoots; crown thinning; branch and crown dieback; reduced growth; yellowing leaves; wilted leaves; leaf and trunk necroses; “bleeding” stripe cankers at root collar; tyloses formation; loosened bark; and sapwood discoloration (reviewed in Jung et al. 1996, Balci and Halmschlager 2002). Tree mortality is gradual (Jung et al. 1996). Reliable diagnosis of oak infection by *P. quercina* in the field is not possible.

Known Hosts

Like other Group I *Phytophthoras*, *P. quercina* appears to have a fairly restricted host range, as only infections of *Quercus* spp. have been reported (Jung et al. 1999).

Hosts	**References**
Quercus cerris (European turkey oak)	reviewed in Cree (2005)
Quercus frainetto (Italian oak)	(Balci and Halmschlager 2002)
Quercus hartwissiana (Hartwissiana oak)	(Balci and Halmschlager 2002)
Quercus ilex (holly oak)	reviewed in Cree (2005)
Quercus petraea (sessile oak)	(Balci and Halmschlager 2003b)
Quercus pubescens (downy oak)	(reviewed in Cree (2005))(Balci and Halmschlager 2003b)
Quercus robur (common, pedunculate oak)	(reviewed in Cree (2005))(Balci and Halmschlager 2003b)
Quercus vulcanica (Kasnak oak)	(Balci and Halmschlager 2002)
Quercus sp. (oak)	reviewed in Cree (2005)

Known Distribution

The pathogen has only been reported from Europe and western Asia.

Location	**References**
Austria	(Balci and Halmschlager 2003c)
England	reviewed in Cree (2005)
France	reviewed in Cree (2005)
Germany	(reviewed in Cree (2005))(Jung et al. 2000, Hartmann and Blank 2002)
Hungary	reviewed in Cree (2005)
Italy	(reviewed in Cree (2005))(Vettraino et al. 2002)
Sweden	(Jonsson et al. 2003, Jonsson et al. 2005)
Turkey	(Balci and Halmschlager 2002)

Potential Distribution within the US

The presence of *P. quercina* has not been confirmed in the US, however a *P. quercina* ‘like’ organism has been identified from oak forests in Minnesota, Wisconsin (Balci et al. 2005) and Missouri (Juzwik, pers. comm.).

A coarse climatic analysis based on reported occurrences of *P. quercina* in the field suggests the pathogen may do particularly well in Mediterranean shrubland (California) and temperate broadleaf forests (northeastern US). Collectively, these two biomes account for approximately 29% of the area within the contiguous US.

Survey

Soil and/or root samples are typically needed to isolate and identify *P. quercina*. Two general approaches are common: direct isolation or baiting techniques (Erwin and Ribeiro 1996). For direct isolation, diseased tissue (cankered bark or necrotic roots) is thoroughly cleaned (rinsed) and surfaced sterilized (e.g., dipped in 95% ethanol). Small pieces are placed on semi-selective media (e.g., PARPNH, an agar medium containing pimaricin, ampicillin, rifampicin, pentachloronitrobenzene, nystatin, and hymexazol). Alternatively, a bait (i.e., a piece of attractive vegetation such as a young oak leaf) is floated on the surface of soil flooded with water. Motile zoospores are attracted to the leaves, cause infection, and cause the bait to decay. Once a bait appears infected, it is rinsed, surface sterilized, and plated just as with the direct isolation method. Occasionally, bacteria or species of *Pythium* may also grow on PARPNH and a number of techniques are available to purify contaminated cultures. Pure cultures are necessary to confirm species identity, based upon morphological or molecular methods.

Jung (1996) collected two to three “soil-root monoliths 20 x 20 x 30cm [9 x 9 x 12 inches]” per tree with a spade. Samples were collected 80-100 cm [31-39 inches] from the base of the tree between root buttresses. Smaller samples were hand dug 50-200 cm [20-80 inches] along main roots. All soil collected from a tree was bulk mixed. Roots were not removed. A 250-500 ml subsample of soil was divided into 30 ml aliquants which were flooded with water to ca. 1-inch depth in 12-cm Petri dishes. Leaflets from *Q. robur* seedling were floated on the water surface. Flooded soils with baits were held at 20°C [68°F]. When a leaf turned blackish brown, the bait was rinsed in demineralized water and dipped in 95% ethanol. Pieces of infected tissue were excised under sterile conditions and placed on PARPNH. If no *Phytophthora* were isolated, the flood water was decanted, and the soil was allowed to dry completely at room temperature. Soils were flooded again and re-baited. The same protocol was used by Jung (2000), and Jonsson et al. (2003).

A similar protocol was used by Hansen and Delatour (1999); however, they removed leaf litter from the soil surface before collecting a soil sample and processed slightly different volumes of soil. A total of ~1L of soil was collected from 4 locations around each tree. Soil was bulked, and a 200 ml subsample was flooded with ~ 500ml of water. Ten young (<3 cm [~ 1 inch]) leaflets of *Q. robur* were floated on the water surface and allowed to incubate for 3 days at 18-20°C [64-68°F]. Vettraino (2002) followed a protocol similar to Hansen and Delatour (1999), but noted that samples were collected in the spring and fall.

Baits of apple or pear fruit did not attract *P. quercina* (Jung et al. 1996).

Species are traditionally identified by measuring and comparing morphological features (including colony growth and spore dimensions) of pure cultures with reported descriptions of known species. Molecular methods for species identification are being developed. Schubert (1999) developed a species specific primer that produced a unique, 1105 bp amplicon for *P. quercina* through polymerase chain reaction.

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Waterhouse, G. M. 1970. The genus *Phytophthora* de Bary. Mycological Papers 122: 101 pp.

Raffaelea quercivora[*]

Scientific Name

Raffaelea quercivora Kubono & Shin. Ito

Synonyms:
none known

Common Names

Japanese oak wilt

Type of Pest

Fungus

Taxonomic Position

Kingdom: Fungi, **Phylum:** Ascomycota, **Order:** Ophiostomatales, **Family:** Ophiostomataceae

Reason for inclusion in manual

Severe plant pathogen vectored by *Platypus quercivorus*, a CAPS Priority Pest (FY 2007)

Pest Description

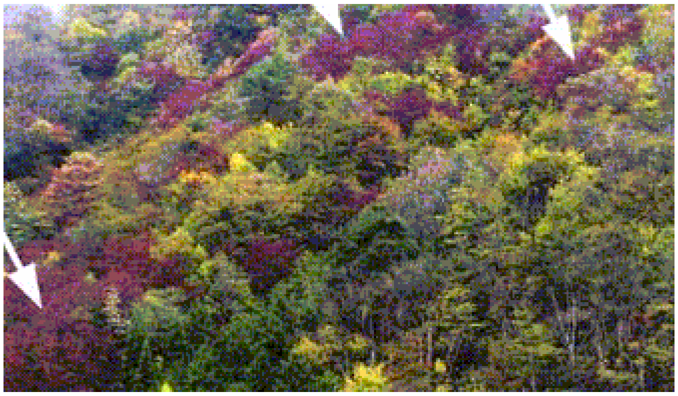

Fig. 1. White arrows point to oak (*Quercus serrata*) wilting due to *Raffaelea quercivora* in Kyoto Prefecture, Japan.
[Image courtesy of K. Kuroda, http://cse.ffpri.affrc.go.jp/keiko/hp/oakwilting-overview.html]

The genus *Raffaelea* was named in 1965 by von Arx and Hennebert and amended two years later by Batra (1967) for ambrosia fungi that create

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[*] This document is largely excerpted from the report: Kromroy, K.W., and R.C. Venette. 2005. Mini-Risk Assessment: Japanese oak wilt, caused by *Raffaelea quercivora* Kubono & Shin. Ito. pp 340-371 *In* R.C. Venette, E.E. Davis, K.W. Kromroy, and S. French. Exotic Pests that Threaten US Forest and Agriculture: Pest Risk Assessments for the Cooperative Agricultural Pest Survey. USDA Forest Service, Internal Report. Available from R.C. Venette (rvenette@fs.fed.us).

sporodochia (small compact cushion-shaped masses of somatic hyphae covered with conidiophores, Fig. 2). Members of the genus produce terminal conidia acropetally (youngest conidium is at the tip or distal end of the chain) (Ulloa and Hanlin 2000) and sympodially (conidia are produced on a conidiophore that terminates and branches repeatedly, producing a zigzag pattern) (Ulloa and Hanlin 2000). In fungal taxonomy, each species is typified by one specimen (a collection, a culture – dried or otherwise preserved in a metabolically inactive state, microscope slide mount) (Alexopoulus et al. 1996), and a "type species" is the basis for description of a genus. *Raffaelea ambrosiae* is the type species for *Raffaelea* (von Arx and Hennebert 1965).

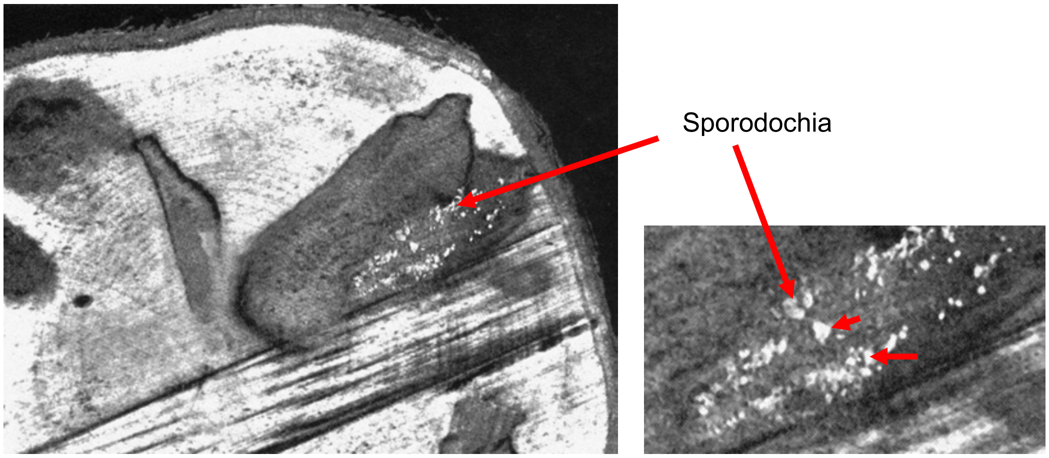

Fig. 2. Arrows point to sporodochia (small white pustules) growing on a cross section of a maple.
[Reproduced from Batra (1967).]

A sexual stage of *Raffaelea* has not been found. Based on numerous similarities to other asexual forms of fungi for which the sexual form is known, *Raffaelea* is classified in the Ascomycetes (Kirk et al. 2001). The morphology and growth habit of spore-producing hyphae (called conidiomata); the shape, septation and color of spores; and the processes involved in conidial production are used to classify asexual fungi (Kirk et al. 2001). Molecular tools provide other evidence of taxonomic identity. Results of a cladistic analysis of 18S ribosomal DNA sequences showed that seven of eight species of *Raffaelea* appear to be phylogenetically related to the genus *Ophiostoma* (Jones and Blackwell 1998). This work occurred prior to the description of *R. quercivora*.

Formal description of the genus *Raffaelea* and type species *R. ambrosiae.* "Sporodochia superficial, effuse, confluent (coalescent), fertile, lush. Conidiophores in fascicles, straight, septate, with a thick base that tapers toward the apex. Conidia originating in succession from the apex of the conidiophore, obovoid, with a conical truncated base, single-celled, hyaline. Aleuriospore solitary, arising from the apex of a septate hypha, globose to even turbinate, single-celled, hyaline, thick covering. *Raffaelea ambrosiae* is the type species, found in *Platypus cylindrus* FAB. in *Quercus*, in England. Type culture in dried agar agaro dessicata CBS 185.64est" (Translated from von Arx and Hennebert (1965)).

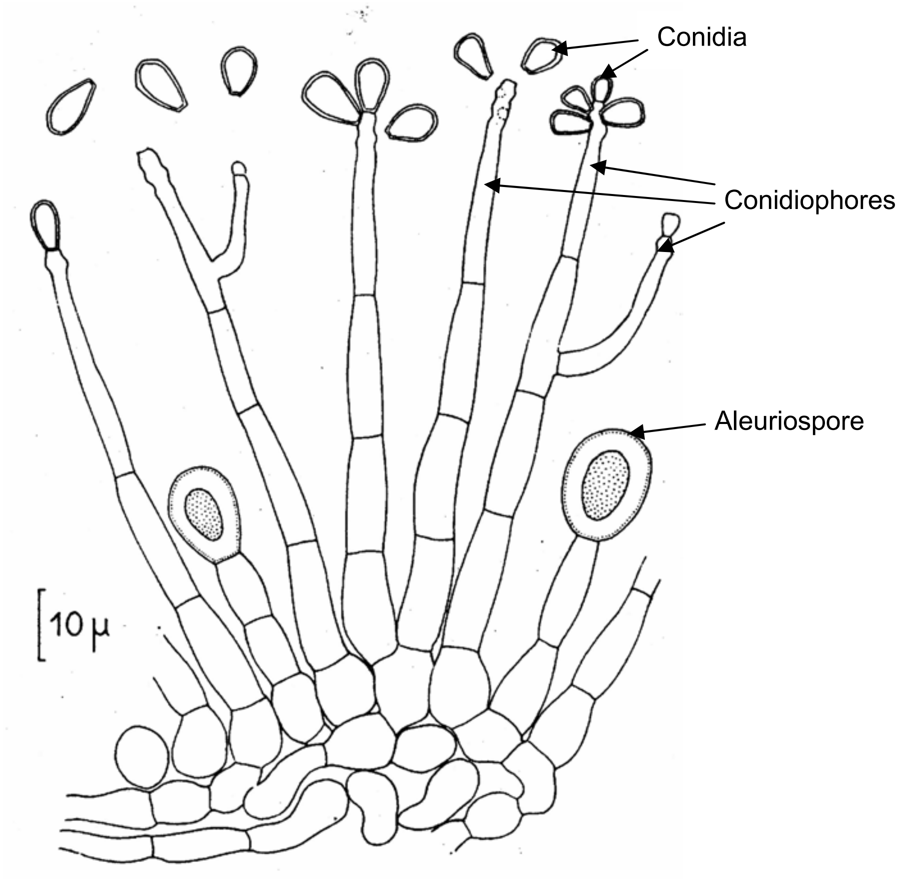

Fig. 3. "*Raffaelea ambrosia*. Conidiophores with conidia and aleuriospores"
[Reproduced from von Arx and Hennebert (1965).]

The original description provided by von Arx and Hennebert was based on examination with a light microscope. In a recent examination of *R. ambrosia* conidial development using scanning electron microscopy, Gebhardt and Oberwinkler (2005) report annellidic percurrent proliferation rather than sympodial production.

Raffaelea quercivora. "Colonies on [potato dextrose agar] PDA at 20–25°C [68-77°F] effuse, spreading rapidly, reaching 80 mm [~3 $^1/_8$ in] diameter in 5 days with an indistinct white margin, appearing water-soaked and mucilaginous; aerial mycelium abundant, floccose, composed of branched, septate, hyaline, smooth hyphae, arranged in fascicles and simulating coremia, reaching 1 cm high; color diffusing and turning pale olive to brown-olive after 2 weeks; odor fragrant, resembling that of ethyl alcohol (Fig. 4). Sporodochia of several fascicles, becoming confluent and mucilaginous. Conidiophores macronematous, mononematous, formed in sporodochia or produced separately, simple or branched, straight, hyaline, septate, smooth, 16.5–22.5 x 0.9–1.5μm (Fig. 5). Conidiogenous cells gradually narrowed toward the apex, polyblastic, sympodial, indeterminate, discrete or integrated, terminal, hyaline, smooth, with a series of flat, scarcely protruding scars situated toward the apex (Figs. 6B and 7C). Conidia short-clavate slimy, borne in acropetal order, hyaline, aseptate, straight, smooth, eguttulate, obovoid to pyriform, tapered markedly toward the truncate base, apex obtuse, frequently yielding sprout cells, formed in droplets, 3.1–4.7 x 2.0–2.4µm" (Figs. 6A and 6C, 7A and 7B)" (Kubono and Ito 2002). Aleuriospores were not described for this species.

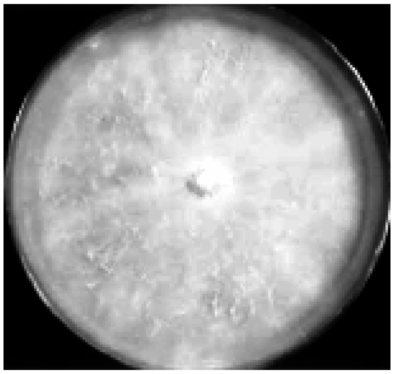

Fig. 4. "*Raffaelea quercivora* colony with sporulation on potato dextrose agar (PDA) at 20º C after 10 days." [Photo and legend from Kubono and Ito (2002)].

Fig. 5. "*Raffaelea quercivora* conidiophores and conidia on PDA." [Photo and legend from Kubono and Ito (2002).]

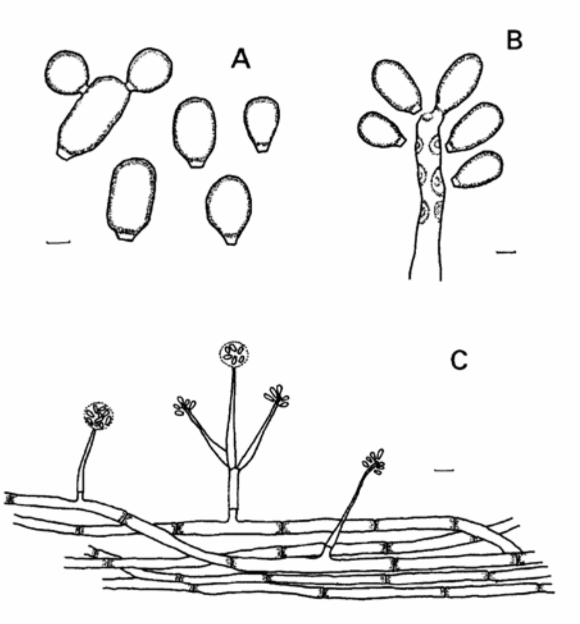

Fig. 6. "*Raffaelea quercivora*. A. Conidia. B. Conidiogenous cell with holoblastic proliferation and cicatricial scars. C. Conidia and conidiophores; one at the left and one at the center showing conidia in droplet. Bars A, B are 1 µm, C is 5 µm."
[Drawings and legend from Kubono and Ito (2002).]

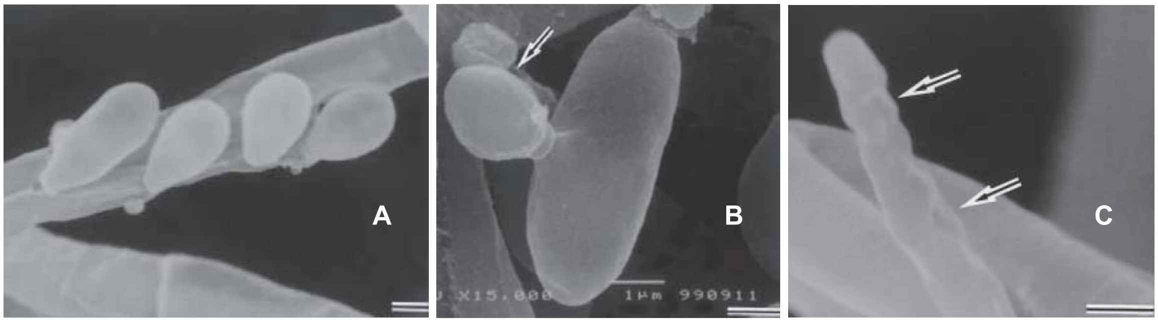

Fig. 7. "*Raffaelea quercivora* on PDA observed by SEM. A. Conidiogenous cell with holoblastic proliferation and conidia. Bar 1µm. B. Conidia and sprout cell (*arrow*) produced from conidia. Bar 1µm. C. Conidiogenous cell with a series of flat cicatricial scars (*arrows*). Bar 10 1µm."
[Photos and legend from Kubono and Ito (2002).]

Biology and Ecology

Raffaelea quercivora is one of many species of ambrosia fungi, so-called because of their symbiotic relationships with some species of wood boring Scolytidae and Platypodidae. Most often associated with recently logged timber and trees of low vitality, ambrosia fungi line the tunnels and galleries created in the sapwood and heartwood by their associated insects with a continuous layer of hyphae and conidia-bearing conidiophores (Batra 1967). The insect relies on the fungus for nourishment during at least some its life stages, meanwhile providing the fungus with protection and means of dispersion and inoculation (Baker 1963, Kinuura 2002).

Similar to other ambrosia fungi, *R. quercivora* has not been found apart from the insect, *Platypus quercivorus* F. (Coleoptera: Platypodidae). Thus, the life cycle of the fungus in nature is intimately related to the life cycle of this insect (Fig. 8). The reciprocal is not necessarily true. Conidia of *R. quercivora* are carried by *P. quercivorus* and contact the plant host when the insect bores into the sapwood and heartwood of the tree (Kinuura 2002). Conidia may germinate directly, or produce sprout cells which germinate and elongate into septate hyphae (Fig. 8A) (Kubono and Ito 2002). Hyphae grow in the tunnels and galleries excavated by the beetle (Fig. 8B), and eventually line the entire surface (Kinuura 2002). Hyphae may aggregate into bundles, called fascicles (Fig. 8C), which may further aggregate into sporodochia (see 'Pest Description') from which conidiophores develop (Fig. 8D) and produce conidia (Fig. 8E). While providing nourishment for the insect, the fungus continues to grow, and hyphae extend into the wood adjacent to the tunnel.

The creation of insect galleries and the presence of the fungus stimulate defense responses from the tree, which include the production of tyloses (parenchyma cell overgrowths) that extend into the xylem and prevent the ascent of water (Yamada and Ichihara 2003). In the meantime, the female insect lays eggs, which hatch in about a week. Larvae feed on the fungus until they pupate. Young adults may emerge in autumn (October, November), allowing for the possibility of a second generation that year, or remain in the tree over the winter and emerge in the spring (Sone et al. 1998). Young adults consume and acquire the fungus in their mycangia. When they emerge from the tree, they carry the fungus. The insects move on to another tree and bring the pathogen to a new host. The fungus may go through many life cycles from the time it is introduced into the tree until the time the next generation of insects emerges with the fungus.

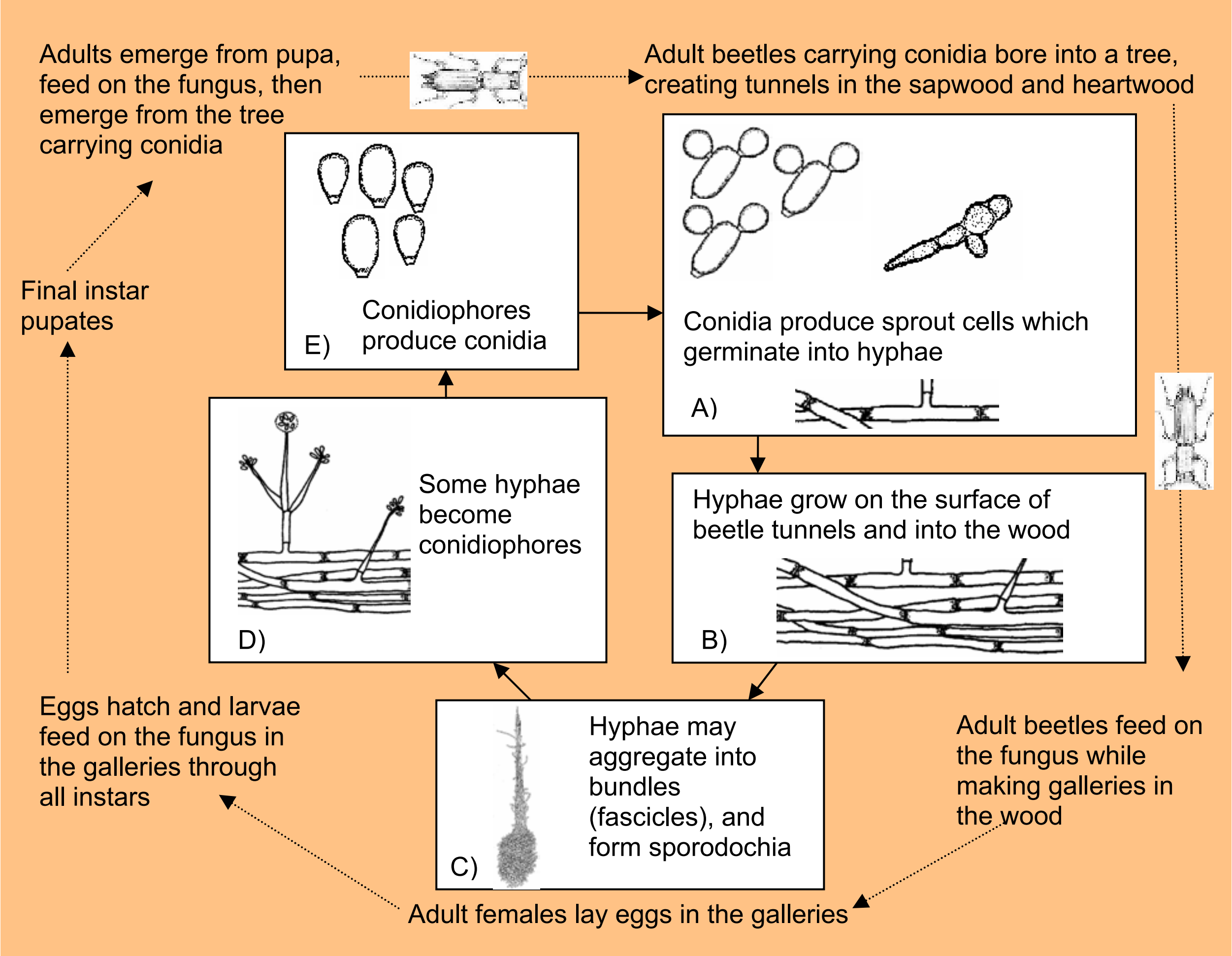

Fig. 8. Life cycle of *Raffaelea quercivora* and its relationship to the vector, *Platypus quercivorus* (images not to scale).
[Drawings from Kubono and Ito (2002), Batra (1967), and Ebeling (1975).]

Infection by the fungus is presumed to occur shortly after initial attack by *P. quercivorus,* when the fungus is first introduced into the host. Attacks start in May or June and may extend through August (Mori et al. 1995, Saito et al. 2001). Following attack by *P. quercivorus*, trees discolor and wilt within 2-3 months and die that first season or by the following spring (Kubono and Ito 2002, Kobayashi and Ueda 2003). Insect infestations of standing trees and logs were highest at the beginning of the season (June – early July) (Mori et al. 1995, Sone et al. 1998).

While there is a tendency to attribute oak mortality to *R. quercivora* when *P. quercivorus* is present, in many cases the fungus was not actually isolated and identified. Spread of the disease within a stand appears to be a function of vector behaviors and patterns. Disease initially occurs at the edge of a gap or forest and on upper slopes; spread among trees occurs outward and downward from an infection epicenter (Kamata et al. 2002, Esaki et al. 2004). Little is known about the likelihood of the fungus being dispersed in the movement of infected logs. Symptoms spread faster in stands that have a higher percentage of susceptible hosts (Kamata et al. 2002).

Specific requirements for the growth of the fungus are not known. In general, nutritional quality of the substrate may determine the primary growth form of *R. quercivora* (e.g. conidia versus hyphae) (Unknown 2002). *Raffaelea quercivora* will grow in culture on PDA at 20-25°C [68-77°F], and produce all of its life stages (Kubono and Ito 2002). In logs, the water content of the wood impacts the growth of *R. quercivora* (Kobayashi et al. 2004).

Hyphae, the filamentous body of the fungus, occur in the tunnels and galleries created by *P. quercivorus*, and will grow into the ray and parenchyma cells of the heartwood. In the process, the fungus stains the wood (Fig. 9). Fragments of hyphae also probably occur in the mycangia and on the outer body surface of the insect vector. On PDA, *R. quercivora* grew to an 80 mm [~3 $^1/_8$ in] diameter colony in five days at 20-25°C [68-77°F] (Kubono and Ito 2002). The fungus has an odor and color on PDA that are distinct from other fungi and yeasts found in *P. quercivorus* mycangia Kinuura (2002). *Raffaelea quercivora* may be tolerant of temperature extremes depending on humidity levels. Hyphae of a related species - *R. ambrosiae* - died after 30 minutes at 50°C [122°F] and 100% relative humidity (RH) but survived 30 minutes of exposure to 75°C [167°F] at 10-20% RH; hyphae continued to grow after 50 days at 2°C [36°F] (Kaarik 1974). Until further research is conducted we can only suggest that tolerances for *R. quercivora* may be similar. For ambrosial symbionts in general, conidia within mycangia are protected by lipids produced by mycangial glands (Baker 1963).

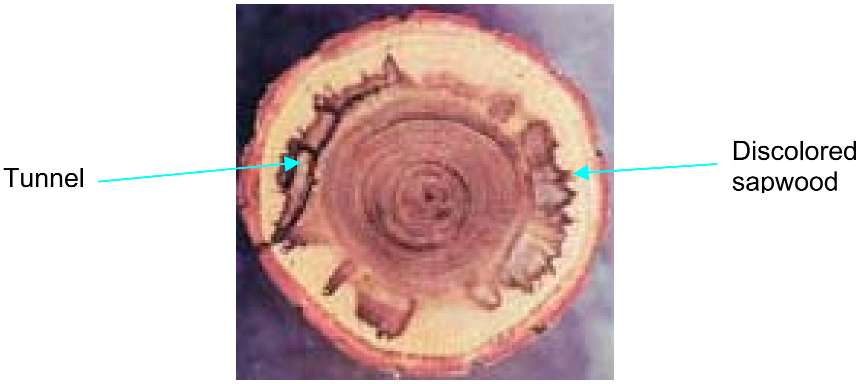

Fig. 9. Cross-section of wilting *Q. mongolica* var. *grosserrata* showing discolored sapwood and insect tunnels.
[Photo from Ito et al. (2003b).]

Wilting results from disruption of water flow at sites of infection (Kuroda et al. 2002, Yamada et al. 2002, Kuroda et al. 2004). Young hyphae of *R. quercivora* invade the living ray parenchyma cells, and tyloses form in the vessels around the hyphae (Kuroda 2001). Tyloses prevent the flow of water and may also form in response to mechanical wounding (Yamada et al. 2002). Trees with significant blockage die, and variation in the amount of blockage may account for differences in mortality rates among and within species (Kamata et al. 2002).

Pest Importance

The complex of *R. quercivora* and *P. quercivorus* has been associated with the mortality of large numbers of oak trees in Japan. Oaks (*Quercus* spp.), mainly *Quercus serrata* and *Q. mongolica* var. *grosseserrata*, are particularly susceptible (Ito et al. 2003b). Since 1980, 100,000 - 200,000 fagaceous trees have been killed annually (Ito et al. 2003b, Ito et al. 2003a). The extensive oak mortality in Japan may have impacted habitat for Asian black bears, causing them to move into more populated areas (Yamazaki 2004). This resulted in numerous human injuries and the killing of numerous bears to stop bear attacks. Approximately 170 bears were killed in one district alone (Yamazaki 2004).

Raffaelea is among the many genera of ambrosia fungi occurring in symbiotic relationships with a group of wood boring scolytid and platypodid ambrosia beetles, but *R. quercivora* is the first ambrosia fungus that has been reported to kill healthy trees (Kamata et al. 2002, Ito et al. 2003b). Oak trees 20-50 cm [ca. 8-20 in.] diameter at breast height (dbh) and 20-30 m [ca. 66-98 ft] tall wilt within 2-3 months after attack by the beetle carrying the fungus (Kubono and Ito 2002). Areas of mortality from Japanese oak disease (Ito et al. 2003b) appear to be expanding (Ito et al. 1998, Kamata et al. 2002, M. Yamato, pers. comm.). The pathogen, which has only recently been described (Kubono and Ito 2002), does not occur in the US at this time and is of concern. The fungus is also a concern in Europe and appears on the European and Mediterranean Plant Protection Organization (EPPO) Alert List (EPPO 2005). The host status of oak species in the US is not known. Nevertheless, given the widespread distribution of oak in this country, especially in the East (Smith et al. 2004), this pathogen poses a serious threat.

At this time, the only known hosts of *R. quercivora* are two species of *Quercus* that occur in Japan. One of these species, *Quercus mongolica*, occurs infrequently in ornamental plantings in the US (USDA 2004), so environmental impacts from insect-pathogen attacks on this species would be minimal. If other *Quercus* spp. in the US are susceptible to attack by *P. quercivorus* and infection by *R. quercivora*, the resulting environmental impacts could be significant.

Symptoms

In the early stages of disease, an *R. quercivora*-infected oak will have curled or withered leaves. Within a few weeks or less, the leaves become discolored and begin to die. By the end of the season or the beginning of the next season, the tree is dead. Currently, *R. quercivora* is only known to be pathogenic on oaks in the white oak group. There are several diseases and abiotic factors that can cause curled or discolored leave. See ‘Survey and Detection’ for a more detailed description of these diseases.

Cross sections of trees infected with *R. quercivora* show galleries of *P. quercivorus* and discoloration that extends beyond the galleries into the surrounding sapwood (Kuroda 1998, Kobayashi et al. 2001). In the US, oak wilt, caused by the well-established fungus *Ceratocystis fagacearum*, is vectored by beetles that do not make extensive galleries in the wood. The pattern of discoloration caused by *C. fagacearum* is different and begins as brown streaks that longitudinally follow the vessels in the outer sapwood (Sinclair et al. 1987, Juzwik et al. 2004). In both diseases, the discoloration in the sapwood becomes darker with time as tyloses plug the vessels.

Known Hosts

Raffaelea quercivora is only known to infect two species within the genus *Quercus*. Only one of these species, *Q. mongolica* var. *grosseserrata*, occurs in the US and only in ornamental settings. Although other oak species are widespread in the US, their host status is not known.

Raffaelea quercivora was identified from samples of discolored sapwood, inner bark and insect galleries of *Quercus serrata* and *Q. mongolica* var. *grosseserrata*. A single, small, host-range test suggests the fungus may be somewhat host-specific; seedlings of six species were inoculated with *R. quercivora*; three were deciduous - *Q. serrata, Q. mongolica var. grosseserrata*, *Q. acutissima*, and three were evergreen – *Q. acuta*, *Q. phyilliyraeoides*, and *Castanopsis cuspidata* var. *sieboldii* (Ito et al. 2003b). *Q. serrata* and *Q. mongolica var. grosseserrata* wilted within 10 days following inoculation, and *R. quercivora* was re-isolated from these trees. Five *Q. mongolica var. grosseserrata* and one *Q. serrata* died by the end of the experiment. No symptoms developed on seedlings of the other four species. Thus, not all *Quercus* spp. are equally susceptible to the pathogen, and the potential host status of oaks in the US is highly uncertain.

Hosts	**References**
Quercus serrata (Japanese common name: ko-nara, bao li) (=*Q. glandulifera*) (=*Q. glandulifera* var. *brevipetiolata*) (=*Q. serrata* var. *brevipetiolata*) (=*Q. urticifolia* var. *brevipetiolata*)	(Kubono and Ito 2002, Yamada et al. 2002, Ito et al. 2003b, CAB 2004)
Q. mongolica var. grosseserrata (mongolian oak, meng gu li, mongori-nara) (=*Quercus crispula*) (=*Quercus grosseserrata*) (=*Quercus mongolica* subsp. *crispula*)	(Kubono and Ito 2002, Yamada et al. 2002, Ito et al. 2003b, CAB 2004, Kuroda et al. 2004)

Several *Quercus* spp. are hosts of other *Raffaelea* spp. A decline of Portuguese cork oak (*Quercus suber* L.) has been associated with outbreaks of *P. cylindrus*, which was found to carry several species of fungi into the trees, including *R. ambrosia* (Sousa et al. 1997). In most cases however, ambrosia fungi have not been associated with symptoms or diseases of trees. Examples of non-pathogenic ambrosia fungi and their vectors found on *Quercus* spp. include: *R. ambrosiae* v. Arx & Hennebert, vectored by *Platypus cylindrus* in England (von Arx and Hennebert 1965); *R. ambrosiae*, vectored by *Platypus compositus* in Arkansas, USA (Batra 1967); *R. tritirachium* Batra, vectored by *Monarthium mali* in Pennsylvania USA (Batra 1967); *R. montetyi,* vectored by *P. cylindrus* in France (Morelet 1998).

Known Distribution

The pathogen has only been reported from Europe and western Asia. *Raffaelea quercivora* has been identified from material collected in several prefectures in Honshu, Japan (Kubono and Ito 2002, Ichihara 2003, Kuroda 2005). *Raffaelea quercivora* may also occur in areas where mortality of Fagaceae, including *Quercus* spp., is associated with *P. quercivorus* attack. Identification of *R. quercivora* has not been reported from these areas, but the symptomatology and frequent presence of unidentified fungi in combination with high numbers of the vector suggest the presence of the pathogen. The distribution of oak mortality appears to be expanding in Ishikawa, Fukui and Shiga Prefectures (Fig. 10) (Kamata et al. 2002).

Location	**References**
Fukui Prefecture, Imajyomachi	(Kubono and Ito 2002)
Fukushima Prefecture	(Ichihara 2003)
Fukushima Prefecture,Takasato Village, Aizubange Cho	(Zhou undated)
Gifu Prefecture, Yanaitsu Cho[1]	(Zhou undated)
Hyogo Prefecture	(Ichihara 2003)
Ishikawa Prefecture[1]	(Ito et al. 1998)
Kii Peninsula	(Ichihara 2003)
Kyoto Prefecture	(Kuroda 2005)
Mie Prefecture	(Ichihara 2003)
Nara Prefecture	(Ichihara 2003)
Nigata Prefecture	(Ichihara 2003)
Ryukyu Islands, Ishigaki Shima island[1]	(Hamaguchi and Goto 2003, CAB 2004)
Shiga Prefecture, Makino-cho (now is Takashima City)	(Kubono and Ito 2002)
Shimane Prefecture	(Ichihara 2003)
Tottori Prefecture, Iwami-cho	(Kubono and Ito 2002)

Location	References
Toyama Prefecture	(Ichihara 2003)
Yamagata Prefecture, Asahi-mura	(Kubono and Ito 2002)

1. These areas are identified as having oak mortality associated with *P. quercivorus* but isolation of *R. quercivora* from these areas was not attempted.

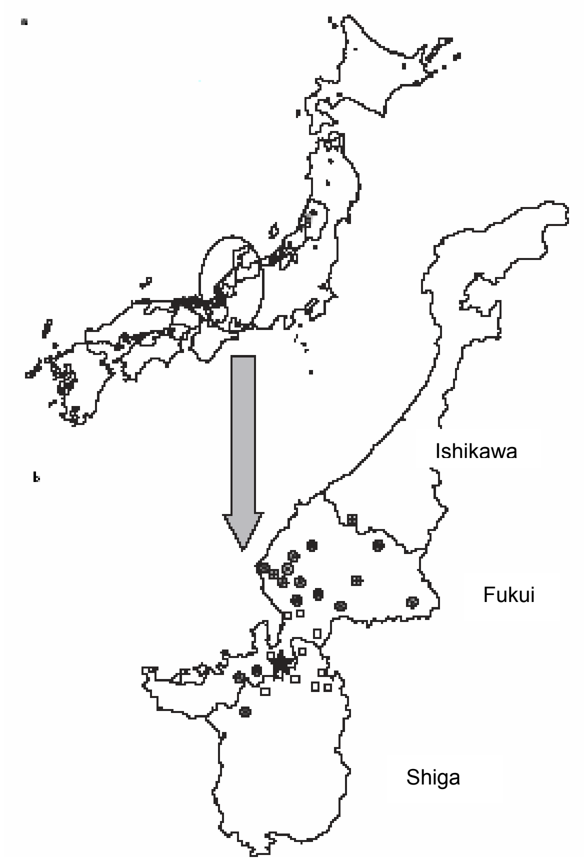

Fig. 10. Map showing occurrence of oak dieback caused by *Raffaelea* sp. 1 (now assumed to be *R. quercivora*) carried by *Platypus quercivorus* in three regions of Honshu, the central island of Japan (after Ito & Yamada, 1998). A detailed map of Ishikawa, Fukui, and Shiga Prefectures, showing localized spread of the disease from a focal point. Symbols denote periods when dieback was observed: ★ < 1980; ☐ 1980s; ◎ 1990s.
[Figure reproduced from Kamata (2002).]

Potential Distribution within the US

Raffaelea quercivora occurs in central Japan, primarily on Honshu. The currently reported distribution of *R. quercivora* suggests that the pathogen may be most closely associated with the temperate-broadleaf-and-mixed-forest biome, generally warm and humid during the growing season but with seasonal temperature and moisture fluctuations. Consequently, we estimate that 28% of the continental US could provide a suitable climate for *R. quercivora*, extending from the northeastern-most states, south into parts of Louisiana and Texas, and west into Michigan, Wisconsin, and Minnesota.

Survey

A survey for *R. quercivora* should target standing oak trees, oak logs and the vector, *Platypus quercivorus*. Methods for monitoring *P. quercivorus* are described in the companion risk assessment by Davis et al. (2005). Because affected hosts and the insect vector may be associated with several species of fungi (Ito et al. 1998, Masuya et al. 1998, Kinuura 2002), identification of *R. quercivora* requires isolation and identification in a laboratory. Aerial surveys using photographs were used to define areas of oak mortality in Japan with some success (Kamata et al. 2001). Landsat imagery was too coarse to accurately identify these areas (Komura et al. 2003).

Only oak species are known hosts of *R. quercivora*, so they should be the focus of a survey. At this time, however, it is unknown whether any oaks in the US are susceptible. Tree crowns should be visually inspected for wilting leaves beginning in June through early September; leaves will appear curled or withered, then become discolored – to a reddish color in Japan (Saito et al. 2001, Kinuura 2002, Ito et al. 2003b, Kobayashi and Ueda 2003). Wilted trees may be dead by August or not until the following spring, and may appear in clusters. In Japan, centers of oak mortality are often found on the edge of a gap or stand (Esaki et al. 2004).

Wilted or dead oaks should be examined for evidence of attack by *P. quercivorus*: entrance holes, most dense within 1 m [ca. 3 ft] of the ground (Hijii et al. 1991), and an accumulation of boring dust and frass at the base (Ichihara 2003). A suspect tree or log should be cut in the field, and one or more cross sections should be examined for galleries of *P. quercivorus* and discoloration that extends beyond the galleries into the surrounding sapwood (Kuroda 1998, Kobayashi et al. 2001). Sapwood discoloration in an infected tree was observed to a height of 4 m [13 ft] (Kuroda 2001). In Japan, sections of the trunk 20-30 cm [ca. 8-12 inches] in diameter and 50 cm [20 inches] long were cut from felled trees or logs, and the ends were coated in the field with a silicone paste to prevent the wood from drying (Kinuura 2002). Sealed logs were taken to a laboratory for further analysis. Logs used as bait for *P. quercivorus* can also be used to detect *R. quercivora* (Kobayashi and Ueda 2003, Kobayashi et al. 2004).

Laboratory diagnosis. *Raffaelea quercivora* can be identified in the laboratory from the sapwood of an infected tree or log and from adult *P. quercivorus*. Log samples were stored at 5°C [41°F], sawed into discs 2-4 cm [ca. 1-2 inches] thick, cut into small blocks and split with a hatchet to expose the insect galleries (Kinuura 2002). For ambrosia fungi in general, Benjamin (2004) suggests preserving samples of thin slices or chips of galleries for later examination, either by drying or by mounting the samples on slides using a fixative mounting medium. Benjamin also suggests aseptically removing slices of galleries and putting them in a sterile moist chamber to encourage further growth of the ambrosia fungus for easier isolation.

A sterile scalpel is used to remove small pieces (e.g., cube 2-3 mm on a side) from the discolored sapwood and insect galleries (Kubono and Ito 2002). Samples are surface disinfected by washing with 80% ethanol and 0.1% solution of mercuric chloride and rinsed in two changes of sterilized water (Kubono and Ito 2002) or by rinsing each cube with 99% ethanol, heating over a flame, and repeating three times (Kinuura 2002). Once disinfected, the cubes are placed on plates of potato dextrose agar (PDA). Bacterial contamination may be inhibited with 100-ppm streptomycin sulphate solution applied to each plate (Kinuura 2002). Areas of discolored sapwood were the areas most consistently associated with isolation of the fungus (Kuroda 2001). It may also be possible to transfer pieces of the fungus directly from the gallery walls and pupal cradles to PDA (Kinuura 2002).

Isolation on PDA and incubation in the dark at 20-25°C [68-77°F] will produce colonies within 5 days (Kubono and Ito 2002). Identification will be based on the size and shape of conidiophores and conidia, and details of conidium production. Identification should be confirmed by scanning electron microscopy (Kubono and Ito 2002, Gebhardt and Oberwinkler 2005). A recent morphological comparison of several species of *Raffaelea* grown in culture highlighted that *R. quercivora* has more slender conidiophores and smaller conidia than other *Raffaelea* spp. (Kubono and Ito 2002).

To attempt isolation of the fungus from the insect vector, *P. quercivorus*, adult beetles should be surface disinfested by immersion in 80% ethanol for 30 seconds, rinsed in a dilute solution of sodium hypochlorite for 2 minutes, then rinsed in sterile distilled water for 30 seconds (Kinuura 2002). Mycangia (Fig. 11) should be excised from female adults using a sharp scalpel or iris scissors and needles. Two or three mycangial sacs are placed on each plate of PDA. Proventriculi (terminal part of the foregut) should be removed from adults of both sexes and placed on PDA (Kinuura 2002). An alternative process is to wash the adults in two changes of sterilized water and place the entire insect directly on PDA (Kubono and Ito 2002). Bacterial contamination may be inhibited with 100-ppm streptomycin sulphate solution applied to each plate (Kinuura 2002). Plates should be incubated in the dark for 2 weeks at 25°C [77°F] or one month at 18°C [64°F] (Kinuura 2002, Kubono and Ito 2002). More than one fungus may be isolated from the beetle (Kinuura 1995). Timing and sex of insect collection can affect the isolation rate of *R. quercivora* (Kinuura 2002).

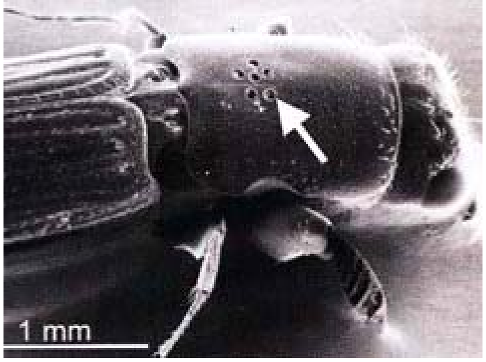

Fig. 11. Female *P. quercivorus* carries conidia in specialized cavities called mycangia (arrow).
[Photo from Kuroda (2005).]

Raffaelea. quercivora will grow rapidly (80 mm [~3 $^{1}/_{8}$ in] diameter in 5 days at 25°C [77°F]), and in 2 weeks will become pale olive to brown olive and have a fragrance (Kubono and Ito 2002). Isolates collected from many different oak trees showed no differences in culture morphology when grown on the same media (Kimura et al. 2005). Microscopic examination of conidiophores and conidia is required for identification, and scanning electron microscopy is better than light microscopy to observe conidiogenesis on fresh culture material (Gebhardt and Oberwinkler 2005). Confirmation should be made by an expert.

Identification of *R. quercivora* requires growth in pure culture. Within the near future, molecular methods may be available as another means of identification (Jones and Blackwell 1998, Kimura et al. 2005).

Similar species. Two diseases that could be confused with the early foliar symptoms of Japanese oak disease are oak anthracnose and bacterial leaf scorch (BLS). Neither anthracnose nor BLS will cause rapid mortality like *R. quercivora*. Anthracnose is caused by the fungus *Apiognomonia errabunda* (imperfect state *Discula umbrinella*), and it occurs on many *Quercus* spp. in the US (Tainter and Baker 1996, Gillman 1999). Spores, spread by rain and wind, infect leaves in spring, causing irregular blotches of dead tissue on leaves. Spots may coalesce and entire leaves may shrivel, turn brown and die. Twigs may also be infected and die. Unlike *R. quercivora* infection, fruiting structures of the fungus may be seen (raised brown flecks) on the underside of lesions (Sinclair et al. 1987). Although unsightly and often recurrent, anthracnose is rarely a serious problem on established trees (Gillman 1999) and outbreaks usually diminish by mid-summer (Sinclair et al. 1987). BLS, caused by the bacterium *Xylella fastidiosa*, is another disease that occurs in oaks across the US, and for which the foliar symptoms may resemble early symptoms of Japanese oak disease (Lashomb et al. 2003, Bentz et al. 2005). The bacterium is transmitted by insects (e.g., leafhoppers) and multiplies in the xylem, eventually blocking water conduction. Leaves show a marginal necrosis or browning, often with a distinct edge bordering the green tissue (Lashomb et al. 2003, Bentz et al. 2005). While symptoms of Japanese oak disease may be apparent early in the season (June) in Japan, BLS symptoms usually appear first in mid to late summer (Lashomb et al. 2003). Symptoms of BLS may occur on only one or a few branches in a season. Over time (years), branches and eventually whole trees may die. Leaves, twigs and branches can be tested for the bacterium by enzyme linked immuno-sorbent assay (ELISA) or polymerase chain reaction (PCR) (Bentz et al. 2005).

A third disease, oak wilt caused by the fungus *Ceratocystis fagacearum,* produces symptoms in red oaks over time that closely resemble those described for infection of *R. quercivora* (Juzwik et al. 2004). *Ceratocystis fagacearum* is vectored by sap and bark beetles and spreads through root grafts with other oaks. Hyphae grow in the xylem and sapwood, stimulate the formation of tyloses and block water conduction. Leaf discoloration and wilting begin in the upper crown within one or two months of infection, which usually occurs in late spring or early summer, and red oaks may be dead by the end of the season. Species in the white oak group are less susceptible, and may decline over many years before dying. Signs on the bark are different for oaks infected with *C. fagacearum* and oaks infected with *R. quercivora*. Red oaks infected with *C. fagacearum* may show small bark crack. If bark is removed, a gray mat of fungal mycelia may be uncovered. These fungal mats grow between the inner bark and the opposing wood, eventually creating enough pressure for the bark to crack (Juzwik et al. 2004). *R. quercivora*-infected trees have no such mats. Descriptions and images of other diseases and problems that produce symptoms similar to the oak wilt caused by *C. fagacearum* should also be reviewed to avoid confusion with disease caused by *R. quercivora* (Juzwik et al. 2004).

No other species of *Raffaelea* have been identified in association with *P. quercivorus*, but several *Raffaelea* spp. have been reported from the galleries and/or mycangia of other ambrosia beetles infecting *Quercus* spp. These fungi include *R. ambrosiae* in England and the US (von Arx and Hennebert 1965), *R. montetyi* in France, (Morelet 1998), and *R. tritirachium* in the US (Batra 1967). These species can be differentiated from *R. quercivora* based on the morphological characteristics outlined in the table below. In general, *R. quercivora* has more slender conidiophores and smaller conidia than other *Raffaelea* spp., having conidia most similar in shape and size to *R. hennebertii* D.B. Scott et J.W. duTiot, vectored by *P. externedentatus* found in *Ficus sycomorus* in South Africa (Scott and Du Toit 1970). The conidiophores of *R. quercivora* are more slender than those of *R. hennebertii* (Kubono and Ito 2002), and the host and location where *R. hennebertii* occurs are very different.

Scanning electron microscopy, a method used to study *R. quercivora* (Kubono and Ito 2002), may be more useful than light microscopy to separate *Raffaelea* spp. based upon morphological characters (Gebhardt and Oberwinkler 2005). Molecular methods will soon be available to separate *R. quercivora* from other *Raffaelea* spp. (Jones and Blackwell 1998, Kimura et al. 2005).

Table 3. Comparison of morphological characters among *Raffaelea* spp.

Species	Sporodochia morphology	Conidiophore (µm)	Conidia (µm)	Conidia shape
Raffaelea quercivora[1]	Fascicles	16.5– 22.5 x 0.9–1.5	3.1–4.7 x 2.0–2.4	Obovoid–pyriform
R. ambrosiae[2]	Superficial	70–150 x 5–9	5–9 x 4–6	Obovoid–triangular
R. montetyi[3]	No description	No description	6.6–13 x 3–6.6	Obovoid–claviform
R. tritirachium[4]	Fascicles	6–15 x 3–5	5–10.4 x 1.5–2.5	Narrowly globose

1. Kubono and Ito (2002)
2. von Arx and Hennebert (1965)
3. Morelet (1998)
4. Batra (1967)

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