July 2005
Perpetual Draft

citrus commodity survey reference

caps

cooperative agriculture pest survey

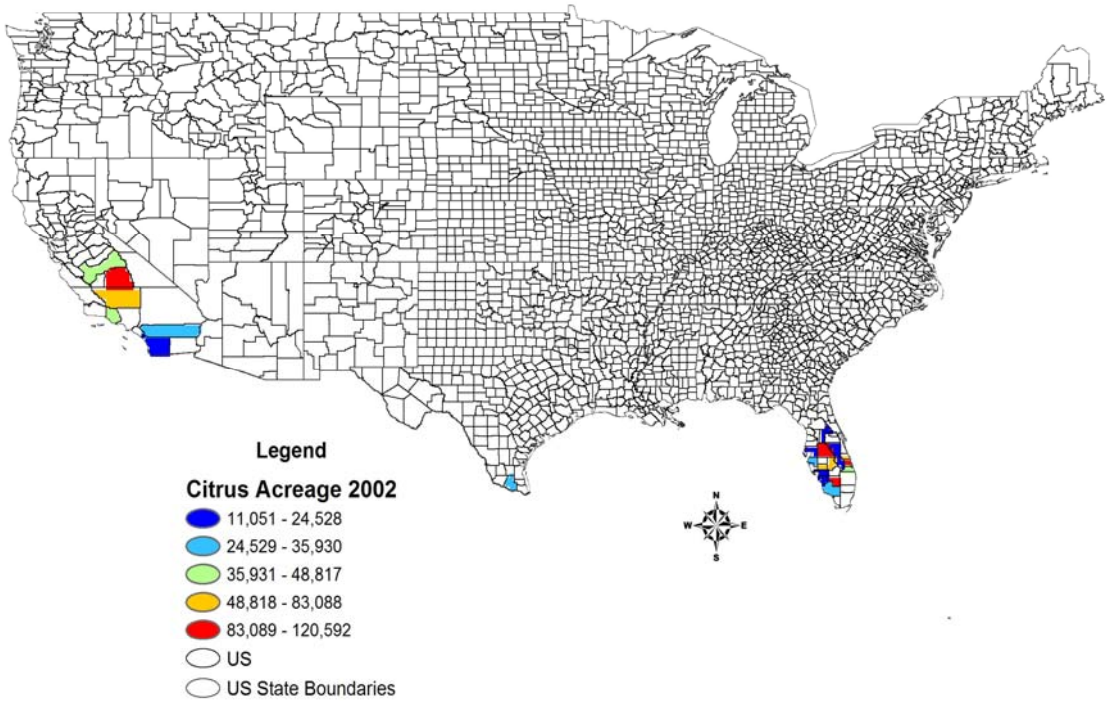
Introduction

Citrus fruits are the most valuable fruit crop in international trade. There are two clearly differentiated markets in the citrus sector: fresh citrus fruits market and processed citrus products market, mainly orange juice. A major development in the last two decades of the twentieth century was the growth in trade of small citrus fruits, (tangerines, clementines, mandarines and satsumas) and a growth in the consumption of citrus fruit juices.

Total annual world citrus production was estimated to be over 105 million tons from 2000 to 2004. Oranges constitute the bulk of citrus fruit production, accounting for more than half of global citrus production in 2004. The rise in citrus production is mainly due to the increase in cultivation areas and the change in consumer preferences towards health, convenience food consumption, and rising incomes.

Citrus fruits are produced worldwide. According to FAO data, in 2004, 140 countries produced citrus fruits. Most citrus fruits, however, were grown in the Northern Hemisphere, accounting for around 70% of total citrus production. The main citrus fruit producing countries include Brazil, the Mediterranean countries, the United States, and China. In the United States, citrus fruits for consumption as fresh fruit are mainly grown in California, Arizona, and Texas; while most orange juice is produced in Florida. These countries represent more than two thirds of global citrus fruit production. The map below illustrates the acreage of citrus grown within the United States. Florida is the leading state for US citrus production. Between 1999 and 2000, the value of fresh citrus shipments in Florida totaled $493.2 million, and the value of processed citrus products was $3,792.2 million.

U S Citrus acreage by county using 2002 NASS data



Citrus Pests

Arthropod Pests

Ants

Solenopsis invicta

Scientific Name
Solenopsis invicta Buren

Synonyms:
Solenopsis interrupta, *S. saevissima* var. *wagneri*, S*. wagneri, S. saevissima richteri*

Common names
Red imported fire ant

Type of Pest
Ant

Taxonomic Position
Class: Insecta, **Order:** Hymenoptera, **Family:** Formicidae

Reason for inclusion in manual
Eastern Region pest list; Regulated plant pest list; and line-item for funding for pest.

Pest Description
The eggs are spherical and creamy-white; the larvae are legless, cream-colored and grub-like with a distinct head capsule. The pupae resemble the worker ants and are initially creamy-white turning darker before the adult ants emerge (eclose). The eggs, larvae and pupae are referred to as a brood (Fig. 1) (CABI, 2004).

Figure 1. Development of *S. invicta* worker ant from eggs (a) larval stages or instars (b-d), pupa (e), and adult (f). Photo courtesy of CABI, 2004.

Worker Ants: The pedicel, or "waist," of the red imported fire ant consists of two segments.The worker ants are wingless, dark reddish-brown with black abdomens, and range in size from 1.5 to 5 mm long (Fig. 2). The mandible has four distinct teeth and the antennae are 10-segmented, ending in a two-segmented club. A sting is present at the tip of the gaster (Collins and Scheffahn, 2003). Worker ants build up the colony, care for the

queen and brood, defend the colony and forage for food. Their function within the colony is determined by the size and needs of the colony, and by the age of the worker ants. The younger workers serve as nurse ants that tend and move the queen and brood. The older workers serve as reserves to defend the colony, and construct and maintain the mound. The oldest worker ants become foragers (CABI, 2004)

Figure 2. Red imported fire ant worker. Photo courtesy of USDA-APHIS-PPQ. www.invasive.org

Sexuals or Winged Reproductives: The females are reddish-brown, whereas the males are shiny and black with a smaller head. These ants stay in the colony until the conditions exist for their nuptial flight. The queen ants are mated female reproductives. They are larger than the worker ants (9 mm) (Fig. 3), and following a nuptual flight will remove their wings (CABI, 2004).

Biology and Ecology (from CABI, 2004)
Solenopsis invicta is a social insect. The colonies usually produce hills, nests, or mounds where they reside. The colonies are most common in open, sunny areas, but occasionally occur indoors and within structures, such as utility housings or tree trunks. In clay-type soils, their mounds may reach 30 to 40 cm high and 30 to 50 cm in diameter. The mounds have no entrance holes or central entrance holes on the surface. The mounds have interconnecting galleries inside, resembling a honey comb in appearance. The mounds may extend 30 to 40 cm deep, although some tunnels can penetrate to the water table. Under extremely hot, dry conditions, the colonies may live underground and not develop surface nests or mounds.

Figure 3. *S. invicta* queen tended by worker ants. Photo courtesy of EPA.

A fully developed colony can contain over 200,000 to 400,000 ants. In response to solar radiation and ambient conditions, the fire ants move within the mound seeking an optimum temperature for the development of the brood (eggs, larvae and pupae), a process called thermoregulation. Foraging worker ants enter and exit through tunnels radiating up to 5 to 10 m away from the mound. The disturbance of mounds results in a rapid defensive response by the worker ants, which quickly run up the vertical surfaces to bite and sting any objects that are encountered.

The worker ant mouthparts are for biting and sipping liquids. The worker ants only consume liquids and particles less than 0.9 microns, storing food in their crops and postpharyngeal gland (oils only) until they feed it to other worker ants and ultimately to the larvae and queen(s). This process is called trophallaxis. The young larval stages (instars) are only fed

regurgitated liquid food. The final (fourth) larval stage can digest solid food particles. The worker ants place bits of solid food in a small depression (called a food basket) just in front of and beneath the larva's mouth (an area called the presternum). They externally digest the proteins (extra-orally) by secreting enzymes, and by chewing and swallowing smaller particles.

The winged reproductive male ants develop from eggs that are not fertilized. The fertilized eggs can develop into either sterile female worker ants or winged reproductive females, depending on the nutrition provided to the larval stages and chemical signals (juvenile hormone level and pheromones) within the colony.

Two forms of *S. invicta* are recognized: monogyne or single-queen colonies and polygyne or multiple-queen colonies. Polygyne colonies contain two or more inseminated, reproductively-active queen ants. The worker ants in single-queen colonies respond defensively to neighboring colonies to maintain territories, whereas multiple-queen colony worker ants do not display territorial behavior. Consequently, the polygene form can produce three to ten times as many ant mounds in a given area of land infested, which can result in 500 mounds and 50 million ants per hectare. In contrast, areas infested with the single queen form normally have 50 to 75 mounds per hectare. The monogyne and polygyne forms can be separated by multiplex PCR of Gp-9 alleles (Valles and Porter, 2003). *Solenopsis invicta* has hybridized with *Solenopsis richteri*, the black imported fire ant, in parts of the southeastern US and produces sexually active offspring (Vinson, 1997).

Ants communicate through vision (sight), vibration (sound), touch and chemicals (pheromones), including a queen pheromone that attracts workers and may suppress dealation, reproduction and a trail pheromone produced by the Dufour's gland that is associated with the worker ant stinger. The worker ants forage when temperatures range from 22°C (72°F) to 36°C (96°F). Upon locating food resources, they develop a trail using a pheromone that directs other worker ants to the site.

Newly mated females that survive nuptual flights and reach suitable nesting habitats (estimated to be about 1% due to predation and other mortality factors), remove their wings and burrow into the ground. They are sealed in a chamber and begin to lay eggs (initially 10 to 20 per day) that hatch into larvae in 6 to 10 days. The larvae are fed from energy produced from the breakdown of flight muscles, infertile (trophic) eggs, young larvae and oil reserves. The first worker ants that emerge are uniformly small and are called nanitics.

Each queen ant can produce approximately 800 eggs per day. The eggs hatch in 8 to 10 days, and the larvae develop through four stages (instars) over 12 to 15 days before pupating for a period of 9 to 16 days. Development requires 22 to 37 days, depending on temperature. Most worker ants live for 60 to 150 days, with the larger ants living longer; during cooler weather the workers can survive for 8 months or more. The newly established colonies develop winged reproductive ants after about 6 to 8 months and can produce 4000 to 6000 alates (sexuals) per year. Queen ants can live for more than 7 years.

Spread occurs through mating flights, ground migration, and floating colonies on water during floods (Lockley, 1996). Mating flights occur between 10.00 h and 14.00 h during the first sunny days after a period of rainy weather when temperature are suitable (24°C), at

any time of the year, but mainly in the spring and autumn. The winged male and female reproductive ants couple in the air (90 to 300 m high). The males die after mating. The females land within a mile or two (unless carried by the wind) and are attracted to shiny surfaces when seeking a suitable nesting site.

Pest Importance

Fire ants are so called because their venom, injected by a stinger like a wasp's, creates a burning sensation. They are also active and aggressive, swarming over anyone or anything that disturbs their nest, including wild animals, domestic animals, pets or people. When a mound is disturbed, ants emerge aggressively to bite and sting the intruder. A white, itching pustule usually appears the next day at the site of the sting (Fig. 4).

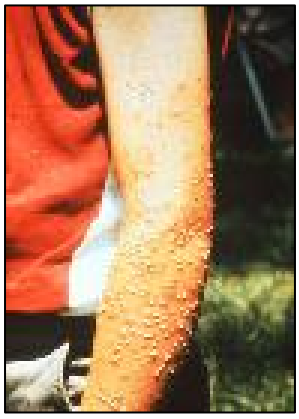


Figure 4. Pustules from fire ant sting. Photo courtesy of T. Lockey.

Fire ants are omnivores that consume sugars (carbohydrates), certain amino acids, ions in solution and some oils containing polyunsaturated fatty acids. Although they primarily consume other arthropods and honeydew produced by some types of sucking insects (Homoptera), they will also consume seeds and other plant parts like developing or ripening fruit, and dead plant and animal tissues. They damage crops, such as soybeans, eggplant, corn, okra, strawberries, and potatoes, by directly feeding on the plants or by protecting other insects that damage the crops. They chew the bark and growing tips of citrus trees and feed on the fruit. Fire ant mounds interfere with farming and mowing operations, turning recreational fields into disfigured moonscapes. Fire ants have caused sections of roads to collapse by removing soil from under the asphalt (CABI, 2004)

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

Figure 5. Fire ant mound. Photo courtesy of CABI, 2004.

Symptoms/Signs

Red imported fire ant mounds are built of soil and are seldom larger than 46 cm (18 in.) in diameter (Fig. 5). The colonies are most common in open, sunny areas, but occasionally occur indoors and within structures, such as utility housings or tree trunks. The mounds have no entrance holes or central entrance hole on the surface (CABI, 2004).

Fire ants damage crops by feeding directly on the plants (Fig. 6A) or by protecting other insects that damage the crops, such as mealybugs (Fig. 6B). They chew the bark and growing tips of citrus trees and feed on the fruit.

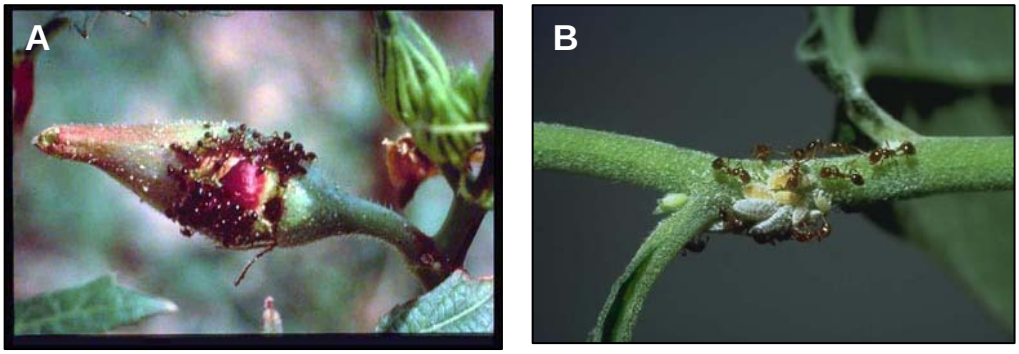


Figure 6. Fire ants feeding on okra bud (A) and tending damaging mealybugs (B). Photos courtesy of B. Ree and B. Dress.

Known Hosts

Solenopsis invicta is omnivorous. Foraging fire ants may be found in or on plants because they are preying on phytophagous arthropods associated with those crops. In addition, fire ant workers feed on certain plants and are attracted to nectaries on plants such as cotton and passionvine. Plant feeding appears to be aggravated by dry or drought conditions. On other plants, the ants seem attracted to oil-containing plant parts, such as the embryo portion of corn and sorghum seeds (CABI, 2004).

Secondary hosts include okra, groundnut, pecan, watermelon, *Citrus* spp., strawberry, soybean, sweet potato, pine trees, nightshade, sorghum, and maize. Red imported fire ants have been associated with cabbage, couch grass (*Cynodon dactylon*), Sickle medick (*Medicago falcate*), buffalograss (*Stenotaphrum secundatum*), and clover (CABI, 2004)

Known Distribution

The red imported fire ant was first introduced from Brazil into either Mobile, AL or Pensacola, FL between 1933 and 1945. The red imported fire ant infests Puerto Rico and eleven of the southeastern states from North Carolina to Texas to Florida; it has recently been found in New Mexico, Arizona, and southern California.

The red imported fire ants are native to Argentina, Brazil, and Paraguay. They have been introduced into Central America (Antigua and Barbuda, Bahamas, British Virgin Islands, Puerto Rico, Trinidad and Tobago, United States Virgin Islands), Queensland Australia, and the United States (Collins and Scheffrahn, 2003; CABI, 2004).

Potential distribution within the US

The red imported fire ants are currently present in eleven states within the United States. Predictions suggest that the ant is unable to survive where the minimum yearly temperatures are less than -12.3°C (10°F) to -17.8°C.

Survey (from CABI, 2004)

Soil that is associated with any articles of trade or shipping equipment from areas known to be infested with *S. invicta* should be carefully inspected. The specimens can be collected in vials of 70% alcohol for preservation and identification by a specialist.

The worker ants of almost all ant species forage away from the colony for food and water at certain times of the day and year. The foraging workers of some ant species, such as fire ants, establish temporary chemical (pheromone) trails that allow nest mates to locate food and water resources. These species can quickly recruit many other ants to a resource. Normally, collected food is brought back to the colony and is fed communally among the other members of the colony, including the queen(s) and brood. Foraging ant surveys can be useful in detecting *S. invicta*. A simple way to survey for fire ant foraging and recruitment is to establish a pattern of 'bait stations'.

Bait stations are made using attractants such as moistened, dry cat food, Vienna sausage pieces, tuna fish or other attractive substances. These are placed in some type of small containers (e.g., scintillation vial or Petri dish) that can be capped upon removal. The vials are placed in a field on their sides in a grid pattern (30 ft to 50 ft grid) or transect line with similar spacing. The bait stations are placed in the field when the ants are foraging, usually when the temperatures range from 18°C (65°F) to 32°C (90°F). The containers are capped after 45 to 60 minutes, and the collected ants are identified and counted. The capped vials can be frozen or filled with alcohol (after the bait is removed) for storage until the sample is analyzed. The ants from each station are identified and counted (or estimated) to provide an indication of the abundance and location of fire ants and other species in the field map.

Counting the number of active fire ant mounds in an area is a simple and easy way to document the population of fire ant colonies. This method assumes that each mound is evidence of a fire ant colony. The results can be used to detect colonies, determine the approach most suitable for the management of ants in the area and monitor the effectiveness of the treatment(s) applied. This approach has several disadvantages, for instance, during, hot, dry periods, fire ants dwell deeper in the soil and do not make a tall, observable mound. Additionally, some fire ant colonies are located in tree stumps, compost piles or other structures where their colony may not be readily observed or associated with a mound. This method does not allow for the detection of native ant species, which may not build mounds at all.

Key Diagnostics

Ants belonging to the genus *Solenopsis* can readily be distinguished from all other ant species in North America by their 10-segmented antennae with a 2-segmented club. These characteristics, combined with the presence of a sting, a two-segmented pedicel and an unarmed propodeum, make identification of the genus relatively easy. Identification of individuals to the species level is somewhat more difficult, especially by the hybridization between two "native" species, as well as between two imported species.

It is important to distinguish between the red imported fire ant and the native fire ant in order to establish appropriate control measures. Mounds of *S. geminata* will contain workers with square-shaped heads that are larger in proportion to the rest of their body. The workers

collect and mill seeds for the colony. Workers of *S. invicta* do not have workers with disproportionate head to body ratios (Collins and Scheffrahn, 2003).

The diagnostic features for *S. invicta*:
The occurrence of fluffy 'worked' soil, particularly a few days after heavy rain. Undisturbed mounds in pastures can reach 18 inches high, but most mounds in turf grass areas are usually just a few inches tall.

The ant mound or nest has no opening in the centre like most ant mounds. Red imported fire ants leave and enter the colony's mound through underground tunnels.

If the mound is disturbed, white objects, which are the eggs, larvae and/or pupae of developing ants are observed.

Fire ants are only approximately one eighth of an inch to a quarter of an inch long. Variation in size is one distinguishing characteristic of imported fire ants. Many other ant species are uniform in size.

When the mound is disturbed, dozens to hundreds of reddish-brown worker ants crawl up the vertical surfaces (like grasses and other objects) on and around the mound. Few native ants do this.

The sting feels like being burned. A day or so later, the imported fire ant's unique venom forms a white fluid-filled pustule or blister at the red sting site, which is characteristic, as only fire ant venom causes this symptom. Worker ants bite (with mandibles) and sting (with stingers) aggressively and repeatedly

Aphids

Toxoptera citricida

Scientific Name
Toxoptera citricida (Kirkaldy)

Synonyms:
Aphis aeglis, *A. citricidus*, *A. nigricans*, *A. tavaresi*, *Myzus citricidus*, *Paratoxoptera argentiniensis*, *Toxoptera aphoides*, *T. citricidus*, *T. tavaresi*

Common Name(s)
Brown citrus aphid, citrus aphid, tropical citrus aphid

Type of Pest
Aphid

Taxonomic Position
Class: Insecta, **Order:** Homoptera, **Family:** Aphididae

Reason for inclusion in manual
Eastern and Western Region pest lists

Pest Description
Worldwide, of the 16 to 20 aphid species reported to regularly feed on citrus, five species are most commonly and consistently encountered in Florida groves: *Toxoptera citricida* (brown citrus aphid); *Aphis spiraecola* (spirea aphid); *Aphis gossypii* (cotton or melon aphid); *Toxoptera aurantii* (black citrus aphid); and *Aphis craccivora* (cowpea aphid). Adult *T. citricida* are shiny-black, and nymphs are grey or reddish-brown (Fig. 1); however, color alone is not distinctive because other aphids on citrus have dark coloration. An additional three species, *Aphis nerii* (oleander aphid), *Macrosiphum euphorbiae* (potato aphid) and *Myzus persicae* (green peach aphid), are rarely collected on citrus in Florida and are not considered pests of the crop (CABI, 2004; Halbert and Brown, 1996).

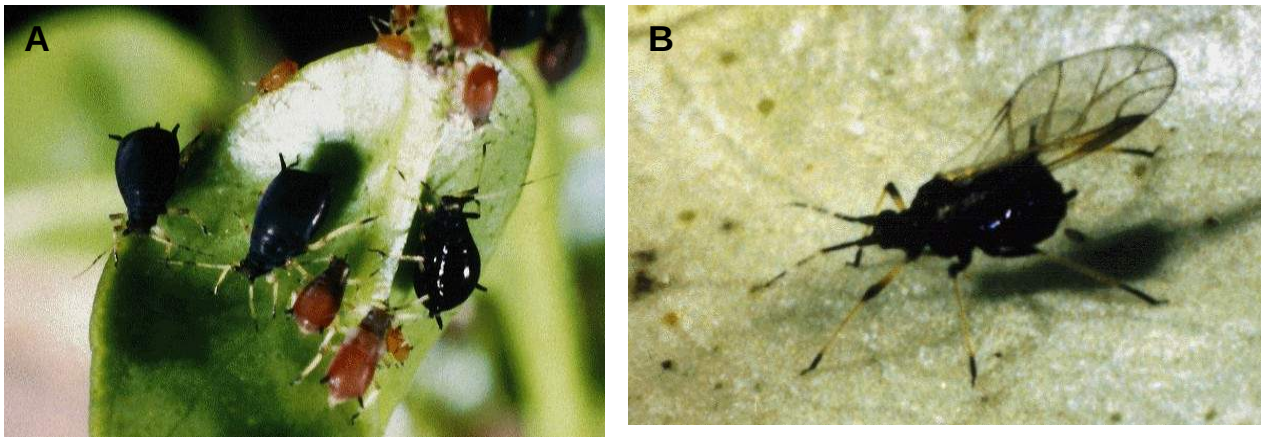


Figure 1. A. Mature apterae (black) and nymphs (reddish brown). **B**. Alatae. Photo courtesy of CABI, 2004.

The brown citrus aphid (BCA) is larger than other species occurring on citrus. Adult wingless forms (apterae) are very shiny black, and nymphs are dark reddish-brown (Fig 1A). Field identification of BCA can be difficult because four of the five regularly collected species can be dark in color, and all five species colonize new citrus growth. Additionally, mixed colonies of two or more species are common. Adult winged forms (alatae) of BCA are distinctive (Fig 1B). They can be recognized by conspicuous black antennal segments I, II and III. Identification is easier with alatae than with adult apterae or nymphs; however, alatae are less common in the field because they tend to leave the colony soon after they emerge (Halbert and Brown, 1996). Halbert and Brown (1996) published a key to adult apterae that will separate most colonies in the field with the aid of a hand lens. Identification of alatae of most aphid species on Florida citrus has been previously published (Denmark, 1990). Stroyan (1961) published another excellent source of identification using microscopic characters. Figure 2 shows common aphid terminology used to distinguish species.

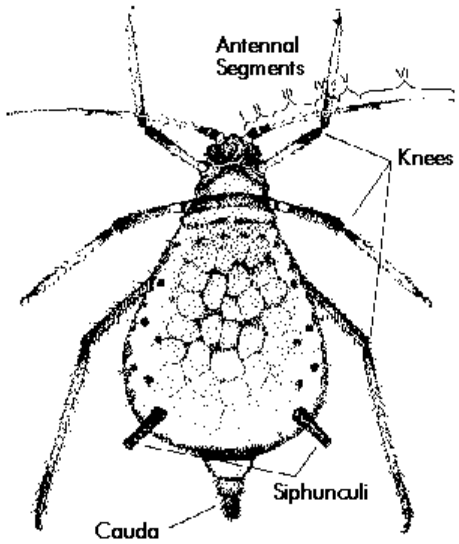


Figure 2: Graphic of wingless form for illustration of aphid terminology useful for field identification. Photo Courtesy of S. Halbert

Winged adult female (alata): 1.1 to 2.6 mm in length; antennae six segmented with I, II, and III heavy black and other segments banded at joints, secondary rhinaria 7 to 20 on III and 0 to 4 on IV, setae on antennae III subequal to or exceeding diameter of segment; siphunculi black, elongate; cauda black, elongate with 25 to 40 setae; stridulatory apparatus on abdomen present; forewing with pterostigma light brown and media usually twice-branched (CABI, 2004).

Wingless adult female (aptera): 1.5 to 2.8 mm in length; oval; antennae six segmented with

no secondary rhinaria; segments not banded, but segments I and II black, segments III and IV pale and slightly swollen, and segments V and VI dark at least at joints, setae on antennae III at least as long as the diameter of the segment; siphunculi black, elongate, and only slightly longer than cauda; cauda black and elongate with about 30 setae; 'knees' of all three pairs of legs very dark; stridulatory apparatus present (CABI, 2004).

Pest Importance

Toxoptera citricida is one of the world's most serious citrus pests. The brown citrus aphid was first discovered in Broward and Dade Counties of Florida in November 1995. Although the aphid alone can cause serious damage to citrus, BCA is a major concern to citrus growers throughout the state because of its high efficiency in transmitting citrus tristeza virus (CTV), a phloem limited closterovirus. Two types of CTV strains are economically important: those that cause the decline of citrus budded onto sour orange (*Citrus aurantium*) rootstock and those that cause stem pitting of grapefruit and sweet orange, regardless of rootstock. Both CTV strains are readily transmissible by *T. citricida*. One of the most devastating citrus crop losses reported followed the introduction of BCA into Brazil and Argentina (when 16 million citrus trees on sour orange rootstock were killed by CTV) (Carver, 1978).

CTV is semipersistently transmitted by citrus aphids. *Toxoptera citricida* is the most important aphid species (of the six) reported to transmit CTV; this is the result of its high vector efficiency, prolific reproduction and dispersal timed with citrus flush cycles to maximize the chances of acquiring and transmitting the virus. Aphids can acquire a virus from infected trees with feeding times as short as 5 to 10 minutes; transmission efficiency increases with feeding times up to 24 hours. There is no latent period, and the virus does not multiply or circulate in the aphid. The time required to inoculate a plant is the same as for acquisition. The aphid is capable of spreading the virus for 24 to 48 hours without re-acquisition. Migrating populations of *T. citricida* are also associated with the spread of certain nonpersistently-transmitted viruses, such as chili veinal mottle virus and soybean mosaic virus in China (CABI, 2004); citrus vein enation (woody gall) virus, a probable luteovirus; cowpea aphid-borne mosaic virus, a potyvirus affecting groundnut in Brazil; and celosea mosaic virus, a potyvirus affecting *Celosea argentea* in Nigeria.

Figure 3. Curled and twisted citrus leaves caused by brown citrus aphid. Photo courtesy of S. Halbert.

Symptoms

New, tender shoots are vulnerable to *T. citricida* colonization and support rapid population build-up. Newly expanding terminals are suitable for BCA growth and reproduction usually for a period of only 3 to 4 weeks, depending on environmental conditions. Aphids are external feeders and extract plant sap from the host by penetrating their stylets into the phloem and injuring citrus by feeding on young growth and causing the leaves to become curled and twisted (Fig. 3). Curled leaves are inefficient energy producers and distortion is permanent. Excess plant sap is excreted as honeydew,

which supports sooty mold growth. This distortion, as well as the sooty mold that grows on the copious amount of honeydew secreted by the aphids, provides excellent protection for scale insects. In addition, sooty mold reduces leaf efficiency and diminishes the quality of the crop. Heavy infestation by *T. citricida* is noted when growing points of citrus are covered by the dark-colored aphids, and the flush bends under the physical weight of the colony. Aphid-tending ants are often present with *T. citricida*, and collect honeydew. When disturbed, *T. citricida* populations sway rapidly in unison, making stridulatory movements with their hind legs, presumably to fend off their enemies. Though flowers are not usually considered a preferred host tissue, high populations of *T. citricida* on flowers have been reported. Mature leaves, stems and fruit cannot sustain *T. citricida* populations.

Known Hosts

Primary hosts of *T. citricida* are citrus and citrus relatives (Family Rutaceae, mostly in the Subfamily Aurantiodeae). Typically, Aurantioideae are trees or shrubs with evergreen leaves. Flowers are usually white and often fragrant. Many genera bear subglobose fruit with a green, yellow, or orange peel with numerous oil glands that result in a nice aroma when handled. Most commercial citrus varieties and rootstocks are good hosts of *T. citricida,* including lime (*Citrus aurantiifolia)*, lemon (*Citrus limon*), pummelo (*Citrus maxima*), tangor (*Citrus nobilis)*, mandarin (*Citrus reticulata)*, tangelo (*Citrus reticulata x paradisi)*, navel orange (*Citrus sinensis)*, satsuma (*Citrus unshiu*), and grapefruit (*Citrus x paradisi*) . In addition, calamondin (x *Citrofortunella microcarpa*), rough lemon (*Citrus jambhiri*), sour orange *(C. auranticum*), box orange (*Severiana buxifolia*) and lime berry (*Triphasia trifolia*) can support *T. citricida*. Orange jessamine (*Murraya paniculata*) is considered a poor host. There are reports that *T. citricid*a have been collected on many non-citrus plants and Barbados cherry (*Malpighia glabra*); however, there is no verification that these are reproductive hosts capable of sustaining an aphid population. These reports may be the result of aphid misidentification (CABI, 2004). Records from potato and sweet peppers may be from plants growing near *Citrus spp* (CABI, 2004).

The aphid may be able to temporarily survive on some non-rutaceous hosts as they migrate away from a crowded food source. Athough Rutaceae are the preferred hosts, large colonies of apterae are often found on different plants, including *Pyracantha* (Rosaceae) in Malawi and Zimbabwe; *Cudramia* (Moraceae) in China and Australia; tea in the Seychelles; and *Maclura* (Moraceae) in Java (CABI, 2004).

Known Distribution

Toxoptera citricida is believed to be native to Asia where citrus originated. Since the first half of the twentieth century, the aphid is widely distributed on citrus in Asia, India, New Zealand, Australia, Pacific Islands (including Hawaii), Africa south of the Sahara, Madagascar, Indian Ocean Islands, South America, and, more recently, Florida. This distribution is attributed to the movement of infested leaves or propagation material. So far, the remainder of U.S. citrus-producing areas and have remained free of the pest. *T. citricida* was found in Spain and Portugal in 2005.

The initial counties found to be infested in Florida were Dade and Broward; the majority of infested trees were in dooryard situations. *T. citricida* now occurs throughout the Florida peninsula.

Potential Distribution Within The US

Toxoptera citricida currently occurs in Florida. Although the aphid is tropical/subtropical in origin, the presence of a sexual stage and overwintering as eggs in Japan suggests that *T. citricida* can adapt to different climates. Due to the restricted host range of the aphid to citrus and its relatives, the most favorable citrus environments for *T. citricida* occur when the weather is warm and humid, resulting in frequent stimulation of new growth cycles. Similarly, desert/semi-arid and cooler regions provide conditions favorable for *T. citricida* seasonally. Populations typically increase rapidly, following colony initiation, resulting in crowding, a decline in host suitability, and production of winged (alate) aphids. Winged morph production could also be triggered by the physiology of the host. A key requirement for the spread of *T. citricida* is that the alata must alight on citrus with new shoot growth to successfully establish a new colony.

Survey

Field infestations of *T. citricida* can be detected by periodic visual inspection of new shoot growth of citrus. Given that the BCA only feeds on tender new citrus terminals, many authors have observed population outbreaks to occur about two weeks following heavy rainfall that induces citrus flush (Michaud, 1998).

Winged forms can be monitored by yellow traps or suction traps. Suction and yellow trap catches often underestimate the number of active BCA colonies in a particular vicinity (Michaud, 1998). Sticky traps and pan traps have been used for monitoring flight activity of the BCA and are more economical than suction traps in terms of capital outlay; however, sticky traps are more attractive to many insects and must be replaced frequently. Furthermore, aphids caught in such traps inevitably require special solvents to be removed and are usually badly damaged, making identification difficult to impossible. Pan traps yield specimens in better condition, but also need to be emptied on a regular basis and are prone to flooding during periods of heavy rain. Traps only monitor the flight activity of alates, providing little information on the survival or location of aphids in citrus groves. Traps are not a substitute for effective survey (e.g., physically searching groves for established colonies).

Alate BCA are not strong fliers and few fly far from the parent colony (Gottwald et al., 1995). Gavarra and Eastop (1976) obtained better catches of BCA in yellow Moericke trays at 152 cm height than they did at trays at ground level. Consequently, optimal placement of traps is probably above ground level, but lower than the height of surrounding trees. Lara et al. (1976) used water traps to compare the attractiveness of various colors to different insects in citrus. In general, they found yellow to be the most attractive to all species, including BCA. Other studies found that the relative attractiveness of yellow and green changed seasonally and varied from year to year (Michaud, 1998).

Key Diagnostics

Toxoptera citricida can be confused with *T. aurantii*, the black citrus aphid, because of its presence on citrus, its dark brown-black coloration, size and the presence of stridulatory apparatus on the abdomen. However, alatae of these aphids can be readily differentiated using a hand lens. *Toxoptera citricida* alatae have antennae III entirely black, forewing pterostigma light brown and media vein lacks the second branch; *T. aurantii* has antennae III, IV, V, and VI banded at joints, forewing pterostigma conspicuously dark blackish-brown

and media vein once-branched. Wingless adults and nymphs are more difficult to distinguish. The easiest character on apterae is the antennae. *Toxoptera aurantii* antennae have several banded joints; whereas *T. citricida* antennae have one prominent band near the middle. Setal length and patterns can be used to differentiate the aphids, but require higher magnification. The cauda of *T. citricida* is bushy with 25 to 40 setae; whereas that of *T. aurantii* is less bushy with 8 to 19 setae. Another black aphid that occurs on citrus is the cowpea aphid (*Aphis craccivora*). It can be distinguished by its strikingly white legs (knees of hind leg may be dark) and 7 caudal setae. For full descriptions and keys of citrus aphids, see Stroyan (1961), Stoezel (1994), and Halbert and Brown (1996) .

Beetles / Weevils

Diaprepes abbreviatus

Scientific Name
Diaprepes abbreviatus Linnaeus

Synonyms:
Curculio abbreviatus, *Diaprepes festivus*, *D. irregularis*, *D. quadrilineatus*, *Exophthalmus abbreviatus*

Common names
Citrus weevil, sugarcane rootstalk borer weevil, West Indian weevil, Apopka weevil

Type of Pest
Weevil

Taxonomic Position
Class: Insecta, **Order:** Coleoptera, **Family:** Curculionidae

Reason for inclusion in manual
Western Region pest list

Pest Description
Eggs: Eggs are smooth, shiny-white in color, oblong to oval in shape, approximately 1.2 mm in length and 0.4 mm in width (Fig. 1). Newly laid eggs are uniformly white, but within 1 or 2 days a clear space appears at either end. Just before hatching, the clear space disappears, the color becomes more brownish, and the mouthparts of the larva are visible within the egg. The number of eggs per cluster varies from 30 to 264 (usually about 60), laid in an irregular cluster composed of a single layer between leaves stuck together with a gummy secretion produced by the female (Woodruff, 1968). This makes the eggs hard to control with conventional insecticides. Eggs are generally laid on new leaf flush. Oviposition occurs from 3 to 7 days after emergence and continues daily for several months. A single female may lay more than 5,000 eggs during her life, which can average 147 days (CABI, 2004).

Figure 1 . Eggs of *D. abbreviatus*. Photo courtesy of R.M. Giblin-Davis.

Larvae: The larvae are white, legless, and 1.5 to 2.5 cm in length (Fig. 2). Its head is creamy to yellow in color; frontale testaceus, testaceus brown towards fore-margin, which is pale to reddish-brown, with a linear border, the tentorial ribs, and a narrow front margin of the parietalia are chestnut brown in color or piceous; parietalia with a narrow pale testaceous streak, which is somewhat broadened anteriorly, on epicranial suture, and a rather faint broad dorso-lateral steak; the latter separated from the testaceus area along frontal suture and an anterior ventral part of parietalia, by pale coloration around setae 1 and 5, which often joins up with the paramedian pale stripe. Sometimes with oblong brownish suffusion on anterior part of paramedian pale stripe; pigmented ocellar spots absent; endocarina absent; anterior margin of sclerotized part of clypeus makedly and evenly raised, forming a transverse ridge or keel; dorso-lateral suffusion of parietalia pale brownish-yellow in color, more or less completely separated from frontal sutures by pale coloration; head width up to 4.11 mm. Skin asperities small, dense and spinulose, especially dense on anterior 5 and last two abdominal segments, on dorsal surface of other segments more or less largely replaced by fine rugosities. Spiracles short-ovate, fringed, with two very small air tubes. Anus 'X'-shaped (CABI, 2004)

Figure 2. Old (left) and young (right) larvae of *D. abbreviatus* larvae.Photo courtesy of P. Grub, USDA.

Pupae: The pupa of *D. abbreviatus* is not described, but that of *D. spengleri* was briefly described by Pierce (1915). It is creamy to white in color, elongate, widest at the mid-point, evenly curved laterally, blunt posteriorly. Each mandible bears large lateral process, which the adult uses to emerge from the pupal cell. Thoracic and abdominal segments each have a transverse row of short setae on small papillae; apical abdominal segment lacking urogomphi. The thoracic spiracle is elongate, and located between the prothorax and mesothorax; abdominal spiracles dark, distinct on segments 1 to 5, very inconspicuous on segments 6 to 8 (CABI, 2004).

Figure 3. The pupal stage of *Diaprepes* can be found in the soil. Photo courtesy of the University of California.

Adults: This is the largest of the weevils found on citrus. Adults measure from 10 to 20 mm in length (Futch and McCoy, 1993).The adults have a black background overlaid by minute white, red orange, and/or yellow scales on the elytra (Fig. 4). The integument is black, except for the legs and antennae, which are dark reddish-brown in color. Antennae are with 2 funicular segment that are very long. The head has a narrow strip of dense, metallic, greenish or pearly white scales at the interior margin of eyes. Laterally from midpoint of eye to well below ventral margin of eye there is a broad band of similar yellowish scales. The rostrum is with median and submedian carinae, declivous apically, and not separated from frons by furrow or distinct impression. The antennal scrobes are flexuous and oblique. Eyes are large, oval, moderately convex. The pronotum is rugose, bearing patches of greenish

Figure 4. Adult *D. abbreviatus* and leaf notching symptom (left) and variation in color and striations of Diaprepes root weevil (right) Photos courtesy of K. Weller and the University of California.

scales, laterally with a broad band of creamy white scales, with vibrissae on anterolateral margin. The scutellum is distinct. Elytra are with distinct shoulders, densely covered with metallic, greenish or pearly scales, tinted with yellow basally on interstices to 4 and along lateral border. Each elytron is with five black, glabrous ridges for half to three-quarters length. Metatibiae are with corbels open, tarsi relatively long, claws paired, free (CABI, 2004).

Biology and Ecology

After 10 to 20 days at 27 to 30 °C, egg masses hatch into neonate grub-like larvae. Newly hatched larvae move across the leaves in a 'peculiar galloping motion' before dropping off from the leaf margins. Larvae usually do not immediately burrow into the ground, but continue to move over the soil surface for several days. The young larvae cannot burrow into dry soil. They eventually fall to the soil surface and move into the soil where they begin feeding on the fibrous feeder roots of the plant. The length of time for larval feeding on the root system varies depending on the species and soil conditions. The larvae complete 10 or 11 instars over a period of 8 to 15 months, attaining a length of 1 inch (2.54 cm). Larval instars 3 through 9 are the most aggressive feeders and may girdle the crown area of the root system, killing the plant. Larvae in the last two instars (10 and 11) feed very little as they enter a quiescent, prepupal period (Grafton-Cardwell et al., 2004).

Prior to pupation, a vertical chamber is formed in the soil at a depth of approximately 45 cm, formed by the larval compaction of the soil by spinning on its caudal end. Pupation occurs within 2 to 3 weeks after the chamber is formed. Newly formed adults remain in the pupal

chamber for at least 11 days, but can remain there for many months before they emerge from the soil. Adult emergence from the soil frequently occurs after a period of extensive rainfall. Adults emerging from pupae are armed with a pair of deciduous mandibles, which break off as they tunnel through the soil. Scars at the site where the deciduous mandibles break off are visible under a microscope. The entire life-cycle, from oviposition to adult emergence, may require from less than 1 year to more than 2 years. In captivity, adults live for several months. Once an adult emerges, it never returns to the soil.

Adults are poor fliers, but can fly from tree to tree. The adult weevil readily feeds on flush citrus and sugarcane leaves. Adult feeding activity occurs during the day and night. Adults can be found throughout the year, with two peak emergence periods occurring between June and September. Heavy rainfall generally precedes peaks in the adult population. The adult males are active for two months while adult females are active for up to four months. They are somewhat social in habit; often, one tree may harbor hundreds, while a nearby tree has none. Mating takes place on the foliage.

Pest Importance

Diaprepes abbreviatus is a root weevil native to the Caribbean where at least 19 additional species of this genus are known. The sugarcane rootstalk borer weevil or "Apopka weevil" was first reported in a Florida nursery near Apopka in 1964. It is one of several species of *Diaprepes* that are major pests of citrus, sugarcane and other crops grown throughout the Caribbean area. It has been observed on more than 75 species of plants in Puerto Rico; it is possible that pests entered Florida on imported ornamentals from this region. Losses attributed to *D. abbreviatus* on Floridaian citrus were estimated at US $73 million during 1995. In Barbados, Metcalf (1959) estimated that larvae were responsible for sugarcane losses of 4.4 tons/ha in first ratoon cane and 2.0 tons/ha in second ratoon crops.

No rootstock appears to be resistant to larval feeding; rootstock tests showed no feeding preference for rough lemon, sour orange, 'Carrizo', 'Milam' or 'Cleopatra'. As few as two weevil larvae per containerized seedling will remove the entire bark from the root system in 4 to 5 weeks. As a result of inadequate management strategies and a wide range of adult and larval food plants, *D. abbreviatus* can be considered a major long-term threat to the survival of several agronomic crops (Simpson et al., 1996).

In Puerto Rico, nursery citrus trees are often affected more severely than mature grove trees (Woodruff, 1968). Jackson (1963) reported that adults fed on female flowers and fruits (Kimri stage) of the date palm, causing the fruit to abort within 48 to 72 hours. The weevil may increase the potential for *Phytophthora* rots to invade the roots of citrus, if present in the soil.

Figure 5. Marginal leaf lotching on citrus. Photo courtesy of Texas A&M University.

Symptoms

Growers might first notice the damage done by the adult weevil feeding on the leaves of citrus plants and become alarmed from the

visual damage to the foliage. However, the unseen damage done by the larvae in the soil, which feed on the plant root system, can have a greater destructive effect on the overall vitality and future productivity of the tree.

The most apparent visual plant damage is a marginal notching of the leaf on young, tender shoots (Fig. 5). Adults feed on the leaves, usually along the edges of new tender leaves, resulting in the characteristic semicircular notching. Unless extensive, feeding by adults is generally considered to have an insignificant affect on citrus and sugarcane. Diaprepes adults leave frass (excrement) scattered on the leaves (Fig. 6A). On rare occasions, adults may feed on fruit of papaya and citrus (Fig. 6B).

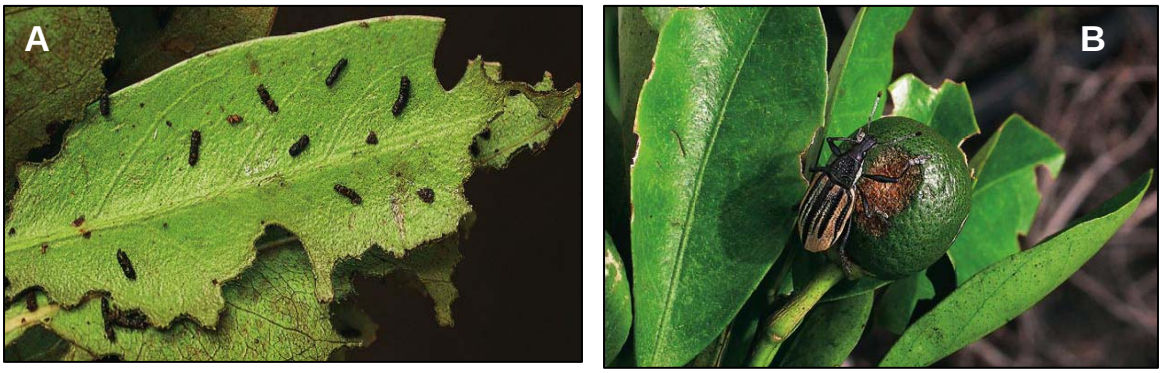


Figure 6. Frass left behind by feeding adults (A) and adult feeding on citrus fruit (B). Photo courtesy of University of California.

Feeding by larvae on roots can result in significant damage to citrus and sugarcane. Injury of sugarcane roots often causes wilting and dieback of the whole plant. In citrus, the roots are eaten by the larvae, causing the plants to become discolored and stunted. Feeding damage by older larvae may be seen on major lateral or pioneer roots when a tree is removed from the soil. Larvae cause damage by channeling on the outer bark tissue into the cambium layer to the woody portion of the root, or by girdling a root, and impeding the uptake of water and nutrients, causing root death. In addition to the damage done to the root itself, channeling on the outer portion of the root could conceivably allow for pathogen invasion. Considerable tree kill has been observed in groves infested with both *Diaprepes* and *Phytophthora* (foot rot). In typical flatwood soils where groves have a shallow root system, larval damage appears to be more pronounced. Since a greater percentage of the root system is close to the soil surface, larval movement through the soil to the roots is much easier.

An infestation of larvae can rapidly kill a young citrus tree. A young tree has a smaller root system and can, therefore, not tolerate the same level of adult and larval feeding damage as a mature tree. Older citrus trees infested by larvae do not always die, but generally become unproductive. Older trees may also be killed if enough larvae are present.

Signs of adult feeding on older leaves, coupled with a thinning appearance of a tree canopy, are good indications that *D. abbreviatus* has attacked a citrus tree. When large citrus trees in Florida, with symptoms of damage by *D. abbreviatus,* have been excavated, large

feeding channels along the bark and into the cambium layer and woody portion of major lateral roots have been observed.

Known Hosts

Adult weevils can feed on a wide range of plants, attacking about 270 different plants. Some of the more common hosts are citrus (all varieties), *Mannihot esculenta* (cassava), *Nephelium lappaceum* (rambutan), peanut, sorghum, guinea corn, corn, Surinam cherry, vegetables, potatoes, papaya, guava, dragon tree, sweet potato, strawberries, sugarcane, panicum grasses, coffee weed (sesbania), Brazilian pepper, woody field grown ornamentals, containerized ornamentals and non-cultivated wild plants (CABI, 2004; Grafton-Cardwell et al., 2004)

Known Distribution

This weevil is present in the Netherlands, the United States (Puerto Rico, Florida, Mississippi, and Texas), Antigua and Barbuda, Barbados, Dominican Republic, Grenada, Guadeloupe, Haiti, Jamaica, Martinique, Montserrat, Puerto Rico, Saint Kitts and Nevis, Saint Lucia, Saint Vincent and the Grenadines, Trinidad and Tobago, and French Guiana.

Potential distribution within the US

Diaprepes abbreviatus was found in Florida in 1964, almost exclusively on citrus.
The pest is mainly distributed in Lake and Orange counties, with smaller infestations in St. Lucie, Palm Beach, and Polk counties.

Survey

The best way to determine the presence of weevils in a citrus grove is to look for the typical notching along the margin of the leaves, particularly young, tender shoots. If notching is present, examine foliage for adult weevils and/or egg masses. Adult weevils feed at night, early morning, or late afternoon, generally hiding within the tree canopy during the heat of the day. Adults feed on a wide variety of host plants year-round, but their peak times are June and September.

Eggs can only be detected through the manual inspection of leaves. Examine the leaves of citrus and sugarcane for notches eaten from the leaf margin. Dig up young citrus and sugarcane plants, especially those showing signs of wilting, discoloration or stunting, and examine the roots for large (up to 2.5 cm), creamy-white, legless larvae, with brown heads or pupae.

Figure 7. A Tedders' ground trap. Photo courtesy of the University of California.

Sour orange (*Citrus aurantium)* seedlings grown in greenhouse pots have 1.8 times as much of the element Rubidium in their leaves as when larvae were feeding on their roots. Manually inflicted damage to the roots simulate weevil damageand have a similar effect. It is suggested that Rubidium uptake could be used to detect root damage as a non-destructive substitute for visual inspection of the roots (Wutscher and Schroeder, 1987).

During the day, trees may be shaken and the ground examined for black weevils, up to 20 mm in length, with red to yellow scales

on the wing covers. The trees on which feeding damage is seen are the trees that should be sampled. A light-colored sheet or canvas below trees will facilitate observation of adults. Shaking foliage after heavy rain will not be as successful, as adult weevils will usually be knocked off the leaves. When the adults are disturbed they will usually fall to the ground faking death.

An inverted funnel trap hung in trees has proven somewhat successful in trapping adults. Researchers often monitor for adults emerging from soil using screened traps; adults are captured as they crawl out of the soil. Recent advances in detecting adults as they emerge from pupating in the soil have been made using a modified version of the Tedders' trap (Fig. 7) (Tedders and Ward, 1994). Growers should use a minimum of 50 cages per infested grove location to determine emergence time. Two cages should be placed near the trunk under each tree to be sampled. The cages should be checked on a weekly basis. In most cases, growers will find low numbers of weevils in each cage, rarely finding more than five. When the first weevil is captured in April or May, this signals the beginning of a major emergence period that can continue throughout the summer for most species. These cages can be used year-round. Once the emergence cages are placed in the grove, care should be used when working with machinery so as not to disturb or destroy them. A pheromone or attractant to add to this trap will enhance surveillance of emerging adults. Light traps have not been a successful survey tool for adults (Beavers et al., 1979).

Key Diagnostics

Currently, there are eight described species of root weevils that are known to infect Floridian citrus. Of these species, five can cause damage of economic importance to nurserymen and commercial growers: *Pachnaeus litus*, *Pachnaeus opalus*, *Asynonychus godmani*, *Artipus floridanus*, and *Diaprepes abbreviatus*. Other less important species include *Tanymecus lacaena*, *Epicarerus fermidolosus*, *Myllocerus undatus* and *Parapantomorus fluctuosus*. These and other species can be found in other parts of the world and are a major production problem in the Caribbean region and South America.

Rhynchophorus palmarum

Scientific Name
Rhynchophorus palmarum Linnaeus

Synonyms:
Calandra palmarum, Cordyle barbirostris, Cordyle palmarum, Curculio palmarum, Rhynchophorus cycadis, R. depressus, R. languinosus

Common Names
Palm weevil, giant palm weevil, palm-marrow weevil, South American palm weevil.

Type of Pest
Weevil

Taxonomic Position
Class: Insecta, **Order:** Coleoptera, **Family:** Curculionidae

Reason for inclusion in manual
National pest List and Western Region pest list (Hawaii)

Pest Description
Eggs: The eggs are laid individually, near the apical area of the palm, 1 to 2 mm inside of the soft plant tissue, and are protected by a brown waxy secretion. Eggs are white, 2.5 x 1 mm in size and have rounded ends (Fig. 1A).

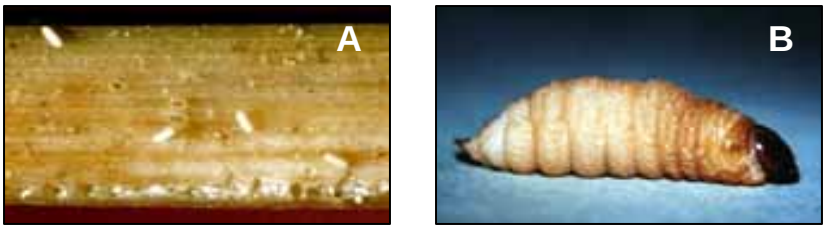


Figure 1. Eggs (A) and larvae (B) of *R. pamarum.* Photo courtesy of CABI, 2004.

Larvae: The larvae are creamy white, legless, initially 3 to 4 mm, and later 5 to 6 cm in length (Fig. 1B). Their body is slightly curved ventrally. They possess sclerotized mouth parts with strong mandibles. Larvae are cannibalistic. Prepupae become darker, and before pupating, they migrate to the periphery of their gallery in the trunk, floral rachis or stem.

Pupae: Pupae are exarate and light brown. If disturbed, the abdomen continuously makes undulatory movements (Fig. 2A). Pupae inhabit a cylindrical-ovoid cocoon, 7 to 9 cm long and 3 to 4 cm in diameter, built with vegetative fibers, organized in a spiral configuration (Fig. 2B).

Adults: Weevils of *R*. *palmarum* have a black, hard cuticle and possess the characteristic elytra of Coleoptera, protecting the abdomen when closed (Fig. 3). They are approximately 4 to 5 cm in length, 1.4 cm wide, and weigh 1.6 to 2 grams. The head is small and round with a characteristically long, ventrally curved rostrum. Adults show sexual dimorphism; the males have a conspicuous batch of hairs on the antero-central dorsal region of the rostrum.

Biology and Ecology

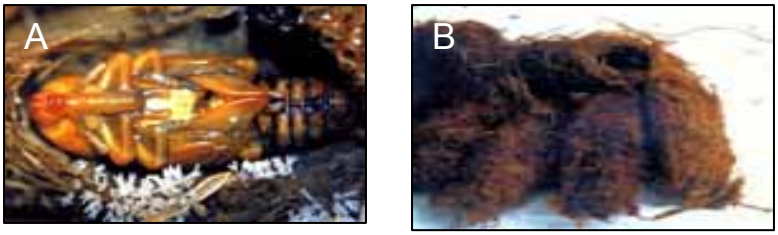


Figure 2. Moulting of *R. palmarum* in pupa (A) and the cocoons of R. palmarum (B). Photo courtesy of CABI, 2004.

The life-cycle of *R*. *palmarum* in the coconut palm is about 80 days (Griffith, 1987) or 120 to 180 days, including 30 to 60 days as adult (Sanchez et al., 1993). The females are attracted to fresh trunk wounds, and lay their eggs inside the plant tissue in a hole made with their rostrum, near or on the internodal area of the palm trunk next to the crown. The larvae of *R*. *palmarum* feed only on live vegetative tissue, and extensively tunnel while developing. Adults are active between 7 to 11 AM, and 5 to 7 PM (Hagley, 1965). They fly only with sunlight, but they avoid flying during the hottest hours of the day (noon and early afternoon). Adults fly at speed of 6 m/s and may travel up to 1.6 kilometers in 24 hours (Griffith, 1987; Hagley, 1965). They respond to attractive odors with a distinct chemotropic and anemotropic behavior. Their preferred habitat is at the base of the leaf axils (Griffith, 1987).

In Central America the maximum adult population occurs during the dry season, and the altitudinal range is from sea level up to 1200 m (Sanchez et al., 1993). Only female adults, measuring 3 cm in size, are the vectors of the nematode *Bursaphelenchus cocophilus*. These females can oviposit 20 eggs in 30 days (Griffith, 1987).

Pest importance

Rhynchophorus palmarum is one of the most important pests on oil palms (*Cocos nucifera* and *Elaeis guineensis*) and ornamental palms (Griffith, 1987; Sánchez et al., 1993). It causes severe damage to palm plantations in Costa Rica, Colombia, Venezuela, and Brazil. A population of 30 larvae can kill an adult coconut palm.

Figure 3. Adult *R. palmarum*. Photo courtesy of Lousiana State Arthropod museum.

In addition to directly damaging plant tissue, *R*. *palmarum* is the vector of the nematode *Bursaphelenchus cocophilus* (Griffith, 1987; Brammer and Crow, 2002). *Bursaphelenchus cocophilus* is the causal agent of the red-ring disease, which causes serious economic losses in palm plantations in South and Central America. Adult female

weevils, which are internally infested with *B. cocophilus,* disperse to a healthy coconut palm and deposit the juvenile stage of the nematode during oviposition. Nematodes enter the wounds, feed, and reproduce in the palm tissues, causing the death of the infected trees. The weevil larvae are parasitized by juveniles of *B. cocophilus,* which persist in the insect through metamorphosis, and appear to aggregate around the genital capsule of the adult weevil. The adult weevils emerge from their cocoons in the rotted palm and disperse to apparently healthy or stressed and dying palms, completing the life-cycle.

Figure 4. Ring ring disease (A) caused by *B. coccophilus* (B), a nematode vectored by *R. palmarum*. Photo courtesy of H Ferris.

Symptoms

Infested palms show a progressive yellowing of the foliage. The emerging leaves are destroyed, and flowers are necrotic. The leaves dry out in ascending order in the crown; and the apical leaf bends and eventually drops. Galleries and damage to leaf-stems made by the larvae are easily detected in heavily infested plants. Tissue of affected plants produces a strong characteristic foul odor. Typically, trees severely attacked by *R. palmarum* show a total death of the palm. Pupae and old larvae are frequently found in the crown area.

If the nematode *B. cocophilus* is present, a crosswise cut of the palm trunk at 0.3 to 2 m above the soil line shows the tell-tale red-ring symptom, which is a circular brick-red area in tall cultivars and usually browner in dwarf and hybrid cultivars (Griffith, 1987). The discolored band is 3 to 6 cm wide, and about 3 to 4 cm from the periphery. Occasionally, in trees older than 20 years, the whole central tissue is red instead of the typical 5 cm band.

Known Hosts

R. palmarum has been reported on 35 plant species (CABI, 2004). The insect is economically important to palms and sugarcane.

Primary hosts: Coconut (*Cocos nucifera*), African oil palm (*Elaeis guineensis*), date palm (*Phoenix dactylifera*), and sugarcane (*Saccharum officinarum*).

Secondary hosts: Avocado (*Persea americana*), cacao (*Theobroma cacao*), banana (*Musa paradisiaca*), mango (*Mangifera indica*), papaya (*Carica papaya*), and pineapple (*Ananas comosus*).

Known Distribution

Central America: Barbados, Belize (widespread), Costa Rica, Cuba, Dominica, Dominican Republic, El Salvador, Grenada, Guadeloupe, Guatemala, Honduras, Martinique, Nicaragua, Panama, Puerto Rico, Saint Lucia, Saint Vincent and the Grenadines, Trinidad and Tobago.

South America: Argentina, Bolivia, Brazil (widespread), Colombia, Ecuador, French Guiana, Guyana, Paraguay, Peru, Suriname, Uruguay, and Venezuela (CABI, 2004).

Potential distribution within the US

Rhynchophorus palmarum and *B. cocophilus* (red–ring disease) have not been found in the USA. It is of great regulatory concern in Florida (Brammer and Crow, 2001). Besides Florida, this weevil could theoretically establish in California, Hawaii, Lousiana, and Texas. Reports of its presence in southeast California and Texas in the past appear to be erroneous (Millar, 2003).

Survey

Detection: *Rhynchophorus palmarum* is only detected when damaged palms start to die. The weevils are usually present at the apical region of the palm crowns. Pupae and old larvae are frequently found when inspecting the crown of infested plants. The palm weevil is attracted to wounds or cuts in the trunks of the palms. Its preferred habitat is at the base of the leaf axils. The presence of a foul odor surrounding the growing points and fruits is another indication of *R. palmarum.* Surveying should be done in the early morning or late afternoon, as adults only fly during the day, although they do avoid the hottest hours. Galleries and damage to leaf-stems made by the larvae are easily visually detected in heavily infested plants. Adults of *R. palmarum* are attracted to palms that have been physically damaged with tools, lightening, or by rats (young palms), in addition to healthy trees. Spear rots or basal rot also attracts the adults.

Trapping: *Rhynchophorus palmarum* is detected by using pheromone baited traps (Oehlschlager et al., 1993). A trap consists of a 4-liter plastic container with windows (15 x 10 cm) for insect entry. Each trap contains a slow release (3 mg/day) pheromone (Rhyncolure) suspended from the lid, and 4 to 5 pieces of halved 10 to 12 cm long sugarcane stalk. These pieces are pre-immersed in 1% a.i. Sevin 80®, (1-naphthyl N-methylcarbamate) or 1% a.i. Furadan®, (2-3-dihydro-2,2-dimethyl-7-benzofuranil methylcarbamate). Pheromone is renewed every 3 to 4 months, and sugarcane pieces every 2 to 3 weeks. Weevils can be removed and counted every two weeks (Alpizar, et al., 2002; Oehlschlager, et al., 2002; Oehlschlager, et al., 1993). Traps can be attached to palm trunks at chest height. Trap density may vary according to stand age, 1 trap/9.5 ha, and 1/6.6 ha, for stands less than 5 years old, and stands 6 to 24 years old, respectively (Oehlschlager, et al., 2002).

Trogoderma granarium

Scientific Name
Trogoderma granarium Everts

Synonyms:
Trogoderma affrum, *T. khapra*

Common Name(s)
Khapra beetle

Type of Pest
Beetle

Taxonomic Position
Class: Insecta, **Order**: Coleoptera, **Family:** Dermestidae

Reason for inclusion in manual
National pest list, Emerging plant pest list, Eastern Region pest list, and Regulated plant pest list

Pest Description
Eggs: Cylindrical, 0.7 mm long and 0.25 mm broad, rounded in one end, and more pointed and with spine-like hairs in the other end.

Larvae: Typically very hairy, yellowish brown to reddish as they mature. They pass through 4 to 7 molts, and are about 6 mm in length and 1.5 mm in breadth when fully mature. The first-instar larva is 1.6 to 1.8 mm long, but more than half consists of a long tail made up of several hairs borne on the last abdominal segment. A characteristic feature of a larva is the presence of single and barbed hairs. Single hairs are scattered over the dorsal surface of the body segments and head (Fig. 1A). Barbed hairs found in pairs of tufts are borne on certain abdominal tergites. In the fourth-instar larva, the hairs give the appearance of four dark transverse bands. *Trogoderma granarium* can be separated from *T*. *versicolor* by the lack or fainting of the dark pretergal line in the 7th abdominal segment, and never present in the 8th abdominal segment.

Pupae: At the last ecdyses, the skin of larva splits, but the pupa remains inside the skin for all its life. Male pupae are smaller (3 mm) than female pupae (5 mm).

Adult: Brown to black oblong-oval beetles, 2 to 3 mm in size, with females being somewhat larger than the males. The head is small and usually deflexed. The dorsal surface is moderately hairy with blurry reddish-brown markings on the wing covers (Fig. 1B). A median ocellus is present between the compound eyes. The number of antennal segments is usually 11, but can be 9.

Figure 1. Adult larva and adult of a *Trogoderma spp.*. Note: Larvae typically very hairy with a brush of long spicisetae like tail. Adult beetles are redish brown with or without vague dark markings, and pronotum dark brown. Photos Courtesy of R. Emery and R. Weinzierl.

Biology and Ecology:
Trogoderma granarium develops most rapidly in hot and humid conditions (18 days at 35°C and 73% relative humidity). Under these conditions, the average number of larval molts is four for males and five for females (Hadaway, 1956). There are two genetic types of *T. granarium* larvae: those that are able to undergo a facultative diapause and those that are unable to do so. Without food, diapausing larvae may survive about 9 months; with food, they may live for 6 years. In this state of very low metabolic activity, they are extremely resistant to the effects of contact insecticides or fumigants. The larvae leave diapause and pupate if subjected to low temperatures for at least a month, and then to warm conditions. A similar, but less effective, stimulus is the introduction of fresh food. Diapause seems to assist survival and dispersal of larva, as diapausing larvae are frequently found on transport equipment, such as sacks and lorries. Pupal development takes about 5 days at 25°C and 3 days at 40°C, and is unaffected by humidity. The virgin female secretes a pheromone to attract an unmated male and, to a lesser extent, mated males. Females only need to mate once. After copulation, oviposition commences immediately at 40°C and lasts 3 to 4 days, while at 25°C; there is a pre-oviposition period of 2 to 3 days. Oviposition may extend over 12 days, and the number of eggs laid is approximately 35 per female. Under optimal conditions, *T. granarium* can sustain a rate of increase of 12.5 times per lunar month. In moister conditions, *T. granarium* is unable to compete against fast-breeding species.

Pest Importance
Trogoderma granarium is one of the most destructive pests of cereal grains and oilseeds. It reduces the weight and grade of the products and makes products unmarketable or unpalatable (Girish et al., 1975). Many countries, including the US, have specific quarantine regulations. The mere presence of *T. granarium* in the US could significantly affect the export of wheat, rice, peanuts, and their products to countries where it is not established.

Symptoms

The first sign of infestation are masses of hairy cast larval skins, which gradually push out from the crevices between sacks (Fig. 2). The larvae crawl over and consume the grain.

Figure 2. Adult beetle, larva, larval skins of *T. granarium*, and damaged wheat kernels Photo courtesy of CABI, 2004.

Known Hosts

The larva of *T. granarium* is a general storage pest found on damaged cereals and oilseeds; to a lesser extent it is found on peas, beans, lentils, and their products. Adults rarely, if ever, eat or drink. Reported hosts: *Arachis hypogaea* (groundnut), *Gossypium* (cotton), *Hordeum vulgare* (barley), *Oryza sativa* (rice), *Panicum miliaceum* (millet), *Sesamum indicum* (sesame), *Sorghum bicolor* (common sorghum), *Triticum aestivum* (wheat), *Zea mays* (maize), *Vigna unguiculata* (cowpea), *Cicer arietinum* (chickpea), *Helianthus annuus* (sunflower), *Pennisetum glaucum* (pearl millet), and *Vicia faba* (broad bean). *Trogoderma* granarium has been intercepted from flowers and fruits of *Citrus limon* and *Citrus sinensis* (Pasek, 2004). *Trogoderma granarium* has been found in non-food sources stored in infested warehouses, transport facilities, and on contaminated materials used for shipping.

Known Distribution

Trogoderma granarium is believed to have originated in India. It is especially prevalent in certain areas of the Middle East, Africa and South Asia. *Trogoderma granarium* does not appear to be established in southeast Asia or Australia. It has been found in South America (Venezuela) and Mexico, but is not established there.

Potential Distribution within the US

In the US, *T. granarium* was found, but later eradicated in Arizona, California, Maryland, Michigan, New Mexico, New Jersey, New York, Pennsylvania, and Texas.

Survey

Inspecting for the Khapra beetle is difficult and meticulous, due to the small size of the insect, its habits, and the difficulty of identifying small or damaged specimens. During inspections, it is most likely to be seen during its larval stage. The diagnostic evidence is the cast of larval skins. Larvae are especially active during the hour before dusk. Special attention should be given to any produce from areas where the pest is known to be widely distributed. *Trogoderma granarium* prefers dark and dry locations, and may remain hidden deep in stored food for relatively long periods. Besides examining products, high risk areas that should be chosen for inspections include 1) cracks in wall and floors 2) behind loose paint or rust 3) grain handling equipment 4) seams and ears of burlap bags 5) low lighted areas 6) trash from cleaning equipment, and the equipment itself. Vacuum cleaners may assist inspectors in drawing cast skins or dead adults from cracks and crevices.

The Khapra beetle is a post-harvest pest. It feeds on dried grains, fruits, spices, and gums. It is only successful in competition with other major stored product pests in conditions of low humidity. It requires hot and dry conditions for at least 4 months of the year (mean temperature greater than 20°C and relative humiditity below 50%)(Banks, 1977).

Key Diagnostics

Populations of *T*. *granarium* may be monitored using commercially available pheromone traps (see: http://ceris.purdue.edu/napis/pests/khb/topics//trap-instruct.html).

Keys for larval and adult identification for *Trogoderma* species of economic importance in North America can be found in Gorham (1987). The confirmation of the identification of *T*. *granarium* may require the dissection of the mouth parts or genitalia (Green, 1979).

Fruit Flies

Anastrepha spp.

Scientific Name (Common Names)
Anastrepha fraterculus (South American fruit fly)
Anastrepha ludens (Mexican fruit fly)
Anastrepha obliqua (West Indian fruit fly)
Anastrepha serpentina (sapodilla fruit fly)
Anastrepha striata (guava fruit fly)
Anastrepha suspensa (caribbean fruit fly)

Type of Pest
Fruit flies

Taxonomic Position
Class: Insecta, **Order:** Diptera, **Family:** Tephritidae

Reason for inclusion in manual
Pest has a line-item for funding and on Regulated plant pest list

Pest Description
The genus *Anastrepha* of frugivorous Tephritidae is endemic to tropical and subtropical regions of the American continent occurring from southern United States to northern Argentina and the Caribbean islands. It comprises about 200 species, distributed into 18 intrageneric groups.

Body: yellow orange to dark brown (Fig. 1).

Head: Higher than long (except in *A. tripunctata*), usually yellow to orange, except for brown ocellar tubercle, but vertex, gena, or occiput sometimes with brown markings, and face rarely with brown or white areas. Frons setulose medially, with 2 to 7 inclinate frontal setae, 1 to 2 reclinate orbital setae, medial and lateral vertical setae, 1 (pair) parallel postocellar, and usually at least 1 small paravertical. Ocellar seta short and much weaker than anterior orbital seta (except well developed in *A. tripunctata*). Postocular setae slender and unicolorous. Face with carina usually well developed, occasionally weak, in lateral view usually straight or concave, but occasionally convex (strongly produced

Figure 1. *Anastrepha* adult females ovipositing on citrus fruit skin. Photo courtesy of CABI, 2004.

medially or dorsally). Antenna short to slightly elongate, usually not extending beyond ventral facial margin; first flagellomere not pointed dorsoapically; arista short pubescent, 1.3 to 1.5 times as long as frons.

Thorax: Mostly yellow orange to dark brown, rarely with dark brown markings; the following areas white or pale yellow, although often not strongly contrasted with darker areas in pale-colored species: postpronotal lobe; dorsal margin of anepisternum; postsutural sublateral vitta from transverse suture to intra-alar seta; usually an unpaired medial vitta, often expanding posteriorly; sometimes a presutural dorsocentral vitta or a presutural lateral vitta (from corner of postpronotal lobe to posterior part of notopleuron); and sometimes apical part of scutellum. Mesonotum completely microtrichose to nonmicrotrichose, sometimes with pattern of bare areas. The following setae well developed: 1 postpronotal, 2 notopleural, 1 presutural and 1 postsutural supra-alar, 1 intra-alar, 1 postalar, 1 dorsocentral, 2 scutellar, and 1-2 anepisternal; acrostichal seta rarely absent; anepimeral seta rarely weak or absent (*A. atrox*); katepisternal seta often weak or absent; dorsocentral seta much closer to level of postalar seta than to level of postsutural supra-alar seta. Scutum, anepisternum and disk of scutellum setulose.

Wing: Occasionally with wasp mimicry pattern with only a complete costal band and a streak in the cubital cells, or rarely with a diffuse pattern, but most species with pattern including 3 bands: a short costal band, or "C-band", ending at the apex of vein R1; an "S-band" formed from a strongly oblique radial-medial band and an anterior apical band, which runs from cell bcu, diagonally across crossvein R-M to join the costa in the apical part of cell r1, and then follows the edge of the wing to the wing apex; and a "V-band", including a subapical band (the "proximal arm" of the V-band), which covers crossvein DM-Cu, and a posterior apical band (the "distal arm"), which crosses cell m, often fusing with the proximal arm in cell r4+5; the C- and S-bands are often connected along vein R4+5, but there is usually a hyaline mark in the radial cells distal to the apex of vein R1, and the S- and V-bands are often connected in cell r2+3. Vein R4+5 setulose dorsally to beyond R-M. Vein M with distal half of the last section curved anteriorly, apex usually strongly curved and meeting costa without distinct angle. Cell bcu with large posteroapical lobe, 2/5 to 1/2 length of basal part of cell. Wing mostly microtrichose, alula, basal cells and hyaline areas adjoining them often partially bare.

Abdomen: Not petiolate.

Male terminalia: Lateral surstylus short (less than height of epandrium), usually flattened. Medial surstylus short, with 2 well-developed prensisetae. Hypandrium apically with laterally compressed hypandrial apodeme. Lateral sclerite long and slender, not connected to hypandrium basally. Phallus elongate, usually 1.25 to 1.50 times length of oviscape in female; reduced in *dentata* and *daciformis* groups. Glans lacking in spp. of *dentata* and *daciformis* groups, in other species elongate, weakly sclerotized, with basal membranous minutely spiculose lobe, and with subapical lobe “T-shaped” and sclerotized.

Female terminalia: Oviscape tube-shaped, often elongate, basally with flangelike lateral lobe. Eversible membrane enlarged basally, this area dorsally with enlarged, usually toothlike scales. Aculeus long and slender, well sclerotized; tip serrate or entire, without large seta-like sensilla (usually with numerous minute and 3 slightly larger sensilla, but none project beyond lateral margin of aculeus). 3 spermathecae.

Pest Importance

Anastrepha is the most economically important fruit fly genus in the American tropics and subtropics. White & Elson-Harris (1992) listed 15 species as significant pests and 28 others that have been reported to attack economically important plants. The worst pest species are the Mexican fruit fly (*A. ludens*), the West Indian fruit fly (*A. obliqua*), the South American fruit fly (*A. fraterculus* (Wiedemann complex)), the Caribbean fruit fly (*A. suspensa*), *A. sororcula*, *A. serpentine*, *A. striata*, the South American cucurbit fruit fly (*A. grandis*), and the Infa fruit fly (*A. distincta*). Several of the species within this genus are illustrated in Fig. 2.

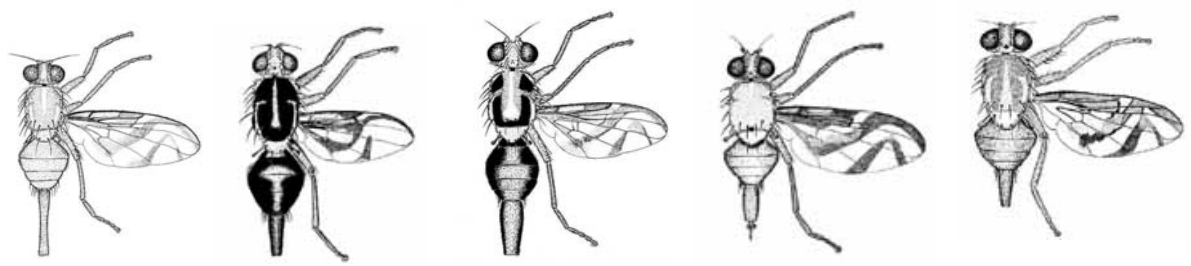


Figure 2. From left to right: *Anastrepha ludens, A. obliqua, A. serpentina, A. striata,* and *A. suspensa*. Photo courtesy of CABI, 2004.

Symptoms

Attacked fruit can show signs of oviposition punctures, but these, or any other symptoms of damage, are often difficult to detect in the early stages of infestation. Much damage may occur inside the fruit before external symptoms are seen, often as networks of tunnels accompanied by rotting. Very sweet fruits may produce a sugary exudate.

Known Hosts

Anastrepha species breed in fruit, either in the pulp and/or the seeds, except for one species (*A. manihoti*), which attacks buds and stems. The host range for the entire genus is very broad, with host plants reported from 143 genera in 54 families; the range is narrower if exotic host plants are excluded. Although there are a generalist species, especially in the *fraterculus* species group, which includes the majority of the pest species, most species individually attack a fairly narrow range of plants. Except for single species in the *serpentina*, *striata*, *leptozona* and *pseudoparallela* groups, feeding on more than a few related hosts or 1 to 2 families by a single *Anastrepha* species is rare outside of the *fraterculus* group. Many species, including the majority of those in the primitive clades, breed in fruits of latex-bearing plants, especially Sapotaceae. Certain species groups are mainly associated with other plant families, such as the *spatulata* group on Euphorbiaceae and Olacaceae, the *pseudoparallela* group on Passifloraceae, the *grandis* group (or at least the one species of the group whose hosts are known) on Cucurbitaceae, the *striata* group on Myrtaceae, and some species of the *mucronota* group on Bombacaceae.

Known Distribution (Potential Distribution within the U.S.)

Currently, *Anastrepha* includes 198 recognized species, as well as numerous other undescribed species. Their combined range includes the southern Nearctic Region (north to

southern Texas and Florida) and all of the Neotropical Region, except Chile, southern Argentina, and several of the Lesser Antilles.

Survey

In most published host plant records for *Anastrepha*, the part of the fruit attacked has not been mentioned. At least in the case of *A. sagittata* and *A. hamata* (*dentata* group) and *A. katiyari* and *A. pallens* (*daciformis* group), the larvae feed exclusively in the seed (Baker *et al*., 1944; Norrbom et al., 1999; Aluja et al., 1999). Larvae of *A. pseudoparallela* (*pseudoparallela* group), and *A. montei* and *A. pickeli* (*spatulata* group) feed on developing seeds and associated tissues similar to species of *Toxotrypana*. Larvae of *A. cordata* (*cryptostrepha* group) mainly feed on the seeds and later on the pulp of the fruit, apparently because immature fruits have large quantities of latex. Larvae of *A. crebra* (*mucronota* group) feed in the pulp and seeds at same time (Hernández-Ortiz & Pérez-Alonso, 1993). Those of *A. steyskali* (*leptozona* group), *A. anomala* and *A. serpentina* (*serpentina* group), and *A. ludens* (*fraterculus* group) can feed on both the seeds and fleshy mesocarp (Stone, 1942; Aluja et al., 1999). Conversely, in most of the generalist species, such as *A. obliqua*, *fraterculus, striata,* and *distincta*, feeding is primarily or exclusively on the mesocarp (Aluja, 1994).

Traps: No male lures have yet been identified for *Anastrepha* spp.; however, they are captured by traps emitting ammonia. Hydrolyzed soya protein and torula yeast were effective for trapping males of *Anastrepha spp.* (CABI, 2004). McPhail traps are usually used for the capture of *Anastrepha* spp. Other baits used for related species include ammonium acetate and casein hydrolysate. IAEA (2003) recommends using a multilure (plastic McPhail) trap with ammonium acetate and putrescine as the attractant with a trap density of 0.25 to 1 trap/km^2 depeding on the area to be sampled (IAEA, 2003).

Key Diagnostics

Most species of *Anastrepha* can be easily distinguished from other Tephritidae by the strongly anteriorly curved apex of vein M, which in most species meets the costa without a distinct angle. In a few species, the curve is not as strong, althougth the distal half of the last section of M is always curved anteriorly. There is overlap in this character with *Toxotrypana*, which differs in having many of the thoracic setae (postpronotal, presutural supra-alar, dorsocentral, acrostichal and scutellar setae) reduced or absent, the scutum with a median longitudinal furrow, and the abdomen petiolate. Additional useful diagnostic characters for *Anastrepha*: Ocellar seta short and weak (except in *A. tripunctata*); dorsocentral seta much closer to level of postalar seta than to level of postsutural supra-alar seta; wing usually with pattern, including C-, S-, and V-bands, although some species have parts of these bands reduced or fused, and others have a wasp mimic pattern with only a complete costal band and a streak in the cubital cells; lateral surstylus short (less than height of epandrium), usually flattened; glans with basal membranous lobe with minute spicules (except in spp. of *dentata* and *daciformis* groups, which lack glans); oviscape tube-shaped, often elongate, basally with flangelike lateral lobe; eversible membrane enlarged basally, this area dorsally with enlarged, toothlike scales; aculeus long and slender, well sclerotized; 3 spermathecae.

Unfortunately, *Anastrepha spp.* are more difficult to identify than most species belonging to the other fruit pest genera of Tephritidae. In particular, it is essential to dissect the aculeus

(ovipositor piercer) of a female specimen to achieve positive identification; a specialist help should be sought in all critical cases.

Bactrocera spp.

Scientific Name (Common Names)
B. correcta (guava fruit fly)
B. cucurbitae (melon fly)
B. dorsalis (Oriental fruit fly)
B. jarvisi (Jarvis' fruit fly)
B. tryoni (Queensland fruit fly)

Type of Pest
Fruit flies

Taxonomic Position
Class: Insecta **Order:** Diptera, **Family:** Tephritidae

Reason for inclusion in manual
Pest has a line-item for funding and on Regulated plant pest list

Pest Description
B. correcta: Similar to *B. zonatus*. It is distinguished by the color of the thorax and the facial spots being united, or almost so, to form a black transverse band. The whitish cross band on the second abdominal segment is less developed. The hind tibiae of the male are distinctly tuberculate before the end. Scutum with anterior supra-alar setae, prescutellar acrostichal setae, two scutellar setae, wing with a reduced pattern (costal band reduced to an apical spot); male with a pecten. The puparium is a dark, shiny yellow. Adults are attracted to open termitaria (Bezzi, 1916).

B. cucurbitae: Pedicel and 1st flagellomere are not longer than ptilinal suture. Face with a dark spot in each antennal furrow; facial spot round to elongate. Frons with 2 to 3 pairs frontal setae; 1 pair orbital setae. Predominant color of scutum is red-brown. Postpronotal (=humeral) lobe entirely pale (yellow or orange). Notopleuron yellow. Scutum with parallel sided lateral postsutural vittae (yellow/orange stripes), which extend anterior to suture and posteriorly to level of the intra-alar setae. Medial vitta present; not extended anterior to suture. Scutellum yellow, except for narrow basal band. Anepisternal stripe not reaching anterior notopleural seta. Yellow marking on both anatergite and katatergite. Postpronotal lobe (humerus) without a seta. Notopleuron with anterior seta. Scutum with or without anterior supra-alar setae; with prescutellar acrostichal setae. Scutellum rarely (5%) with basal as well as apical pair of setae. Wing length 4.2 to 7.1 mm. With a complete costal band; depth to below R2+3, sometimes reaching R4+5. Costal band expanded into a spot at apex, which extends about half way to M. With an anal streak. Cells bc and c colourless. May have a transverse mark over crossvein r-m. Always with transverse mark over crossvein dm-cu. Cells bc and c without extensive covering of microtrichia. Cell br (narrowed part) with extensive covering of microtrichia (White and Hancock, 1997).

B. dorsalis: Belongs to a subgroup that has yellow postpronotal lobes, parallel lateral vittae, and femora not extensively marked. Within this group it is distinguished by its short aculeus/aedeagus; tomentum with no gap; narrow costal band; narrow abdominal markings. *Bactrocera dorsalis* species complex distinguished as follows: *Bactrocera spp.* with a clear wing membrane, except for a narrow costal band (not reaching R4+5); cells bc and c colorless (except in a few non-pests with a very pale tint) and devoid of microtrichia. Scutum mostly black; with lateral but not medial vittae; yellow scutellum, except for basal band which is usually very narrow; abdomen with a medial dark stripe on T3-T5; dark laterally (but form of marking varies from species to species) (Drew and Hancock, 1994).

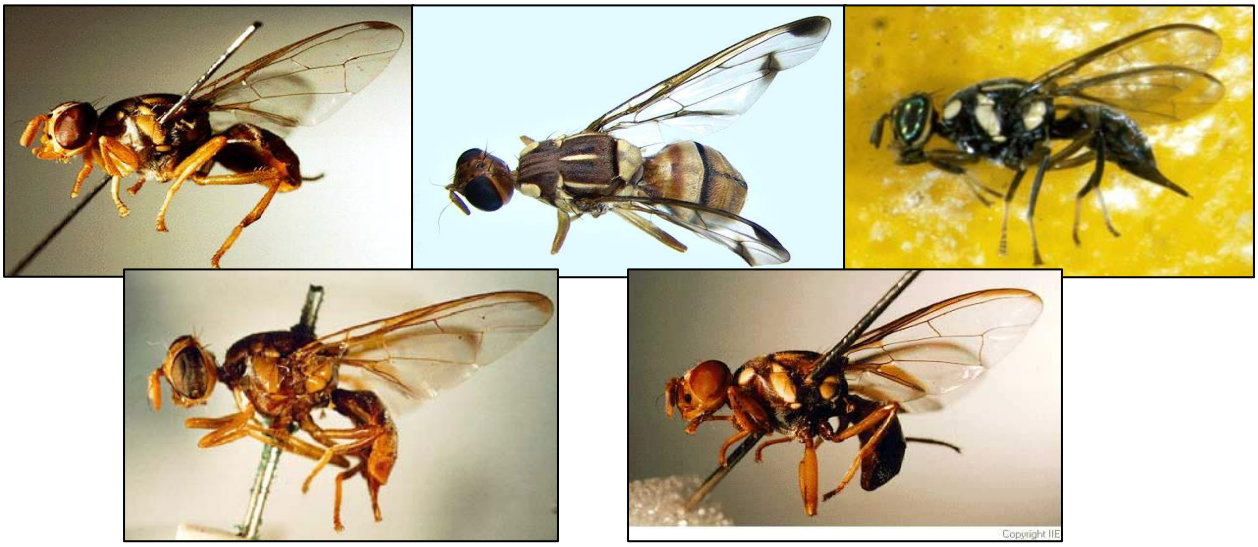


Figure 1. Top (left to right) *B. correcta*, *B. cucurbitae, B. dorsalis*. Bottom (left to right) *B. jarvisi*, and *B. tryoni*. Photos courtesy of CABI, 2004.

B. jarvisi: Has patterned wings, and the female has a long telescopic and pointed ovipositor; these features are hardly known outside the Tephritoidae. The family Tephritidae may also be separated from all other Diptera by the presence of setulae along the dorsal side of vein R1, and the shape of the subcostal vein, which bends abruptly through a right-angle and fades to a fold before reaching the wing edge. At the wing base, *Bactrocera* and *Dacus* species have a very deep cell bm and a very long pointed extension of cell bcu (cup). The genus *Bactrocera,* is separated from *Dacus*, by the terga (dorsal sclerites of the abdomen) not being fused into a single sclerotized plate. *Bactrocera jarvisi* is a very pale colored species. As with most species in or close to the subgenus *Bactrocera*, the scutum has two pale lateral vittae (lateral stripes). The scutum has prescutellar acrostichal setae but not anterior supra-alar setae (the feature that separates subgenus *Afrodacus* from subgenus *Bactrocera*); there are two setae (apical pair) on the margin of the scutellum. An unusual feature is the lateral pale stripes on the prescutum. The males have a deep V-shaped notch in the fifth sternite and a pecten (comb of setae) on each side of the third abdominal tergite (White and Hancock, 1997; Drew, 1989).

B. tryoni: Pedicel+1st flagellomere not longer than ptilinal suture. Face with a dark spot in each antennal furrow; facial spot large, round to elongate. Frons with 2 pairs frontal setae; 1 pair orbital setae. Predominant color of scutum red-brown. Postpronotal lobe entirely pale (yellow or orange). Notopleuron yellow. Scutum with lateral postsutural vittae (yellow/orange

stripes), which do not extend anterior to suture, are tapered, and reach to the posterior supra-alar seta. Scutum without a medial vitta. Scutellum entirely yellow (except for narrow basal band). Anepisternal stripe not reaching anterior notopleural seta. Yellow marking on both anatergite and katatergite. Postpronotal lobe (humerus) without a seta. Notopleuron with anterior seta. Scutum with anterior supra-alar setae and prescutellar acrostichal setae. Scutellum without basal setae. Wing length 4.8 to 6.3 mm. With a complete costal band, which may extend below R2+3, but not to R4+5; not expanded into a spot at apex. With an anal streak. Cells bc and c coloured. No transverse markings. Cell bc without extensive covering of microtrichia. Cell c with extensive covering of microtrichia. Cell br (narrowed part) with extensive covering of microtrichia. All femora yellow/pale (White and Hancock, 1997).

Pest Importance

The genus *Bactrocera* is the most economically significant genus, with about 40 species considered to be important pests (White & Elson-Harris, 1992). Many of them are highly polyphagous. *Bactrocera* is native to the Old World tropics, and most of the major pests are from the Oriental and Australasian Regions. Among the most important species are the Oriental fruit fly (*B. dorsalis*) and several closely related species recently revised by Drew & Hancock (1994), the melon fly (*B. cucurbitae*), the olive fruit fly (*B. oleae*), the Queensland fruit fly (*B. tryoni*), and the peach fruit fly (*B. zonata*).

Figure 2. Ovipositional sites of *Bactocera spp.* Photo courtesy of CABI, 2004.

Symptoms

Adult fruit flies damage the fruit where they lay their eggs causing blemishes and discoloration. Attacked fruit can show signs of oviposition puncture (Fig. 2), but these, or any other symptoms of damage, are often difficult to detect in the early stages of infestation. Following oviposition, there may be some necrosis around the puncture mark ('sting'). This is followed by the decompostion of the fruit.

The maggots bore into the fruit (Fig. 3), develop inside and pave the way for secondary invaders (fungi or bacteria), which cause extensive rotting and fruit drop. Damage can occur inside the fruit, often as networks of tunnels accompanied by rotting, before external symptoms are visible. Very sweet fruits may produce a sugary exudate. Damaged fruits are unfit for human consumption. Damage symptoms do not vary on different crops.

Figure 3. Larvae of *B. tryoni* inside damaged fruit. Photo courtesy of D. Astridge and H. Faye, Queensland Department of Primary Industries

Known Hosts (from CABI, 2004)

Bacterocera spp. have hosts within at least 34 plant families, including Anacardiaceae, Arecaceae, Cucurbitaceae, Ebenaceae, Lecythidaceae, Moraceae, Musaceae, Myrtaceae, Passifloraceae, Punicaceae, Rosaceae, Rubiaceae, Rutaceae, Sapotaceae, and Solanaceae.

Economically important hosts include cashew nut, mango, guava, *Citrus* spp., melon, squash, cucumber, tomato, passionfruit, bean, fig, avocado, cowpea, bell pepper, persimmon, coffee, apple, banana, apricot, peach, plum, pomegranate, pear, kumquat, pumpkin, walnut, and grape.

Known Distributions (Potential Distributions within the US)

Bacterocera correcta has been recorded in China, India, Myanmar, Nepal, Pakistan, Sri Lanka, and Thailnd (CABI, 2004; Hancock, 1991; White and Elson-Harris, 1992). A few individuals were trapped in California during 1986, but these adventive populations have not become established (Weems, 1987).

Bacterocera cucurbitae has been reported in Afghanistan, Bangladesh, Brunei Durussalam, Cambodia, China, India, Indonesia, Iran, Laos, Malaysia, Myanmar, Nepal, Oman, Pakistan, Phillipines, Saudi Arabia, Singapore, Sri Lanka, Thailand, United Arab Emirates, Vietnam, Cameroon, Egypt, Gambia, Guinea, Kenya, Mali, Mauritius, Reunion, Seychelles, Somalia, Tanzania,United States (Hawaii), Guam, Kiribati, Nauru, Northern Mariana Islands, Papua New Guinea, Soloman Islands, and Australia. The Asian species distribution represents its natural (native) range. This species arrived in Hawaii in late in the 19th century (Clausen, 1978). Old records from Darwin, Australia indicate an eradicated outbreak cerca 1910; however, as no specimens were available, this may indicate a misidentification of *Bactrocera chorista*.

Bacterocera dorsalis has been reported in Bangladesh, Bhutan, Cambodia, China, India, Indonesia, Laos, Myanmar, Nepal, Pakistan, Sri Lanka, Thailand, United Arab Emirates, Vietnam, Belau, French Polynesia, Gaum, Northern Mariana Islands and the United States. The recent revision by Drew and Hancock (1994) has shown that the “true” *B. dorsalis* is restricted to mainland Asia (except the peninsula of southern Thailand and West Malaysia), Taiwan and its adventive population in Hawaii. EPPO (2003) also includes California and Florida in its distribution of *B. dorsalis* because the fly has been found to be repeatedly trapped in these regions in small numbers.

Bacterocera jarvisi has been reported in Australian Northern Territory, New South Wales, Queensland, and Western Australia (Hancock et al., 2000).

Bacterocera tryoni has been reported in Queensland, New South Wales, Victoria, Western Australia, and South Australia. A few males have been trapped in Papua New Guinea, but it is unlikely to become established there (Drew, 1989). It is also adventive in French Polynesia (Austral and Society Islands), New Caledonia and twice in Easter Island, although it had been eradicated both times (Bateman, 1982). EPPO (2004) reports that this species was formerly present in California.

Survey

Fruits (locally grown or samples of fruit imports) should be inspected for puncture marks and any associated necrosis. Suspect fruits should be cut open and checked for larvae. Larval identification is difficult, so if time allows, mature larvae should be transferred to sawdust (or a similar dry medium) to allow pupariation. Upon emergence, adult flies must

be fed with sugar and water for several days to allow hardening and full color to develop before they can be identified.

Traps: Males of most *Bactrocera* spp. (including *B. correcta*, *B. cucurbitae*, and *B. dorsalis*) are highly attracted to methyl eugenol (4-allyl-1,2-dimethoxybenzene) and the cue lure (4-(p-acetoxyphenyl)-2-butanone). Many countries that are free of *Bactrocera* spp., for example, the United States (particularly California and Florida) and New Zealand, maintain a grid of methyl eugenol and cue lure traps in high risk areas (ports and airports), if not around the entire climatically suitable area. The traps are usually modeled on the Steiner trap (White and Elson-Harris, 1994). Although these two para-pheromones (methyl eugonl and cue) are considered to be highly volatile, they can be used with panel, delta, or bucket traps. Males of *B. tryoni* are attracted to cue lure, sometimes in very large numbers (IAEA, 2003; CABI, 2004).

Mohammed Jalaluddin (1996) devised a modified new trap that was more efficient at attracting *B. correcta* males than a conventional model. The frequency of distribution of *B. correcta* catches per trap per day, in different entrance port sizes (5 to 30 mm diameter), varied. Orange and yellow colored traps attracted more fruit flies, recording 4.34 and 4.18 numbers/day respectively, compared with green, red, white, violet and blue. Traps placed at heights approximately 1.5 and 2.1 m caught the highest numbers of *B. correcta*. Traps placed at the orchard's border intercepted a greater number of invading adults, more of which were male. Twelve new attractant traps with methyl eugenol were hung by nylon fishing line. Each week, trapped files were collected and counted; fresh bait was then added. Since the sex ratio is 1:1, the population can be intercepted and monitored; timely control measures can be applied.

Bactrocera jarvisi is a particularly difficult species to detect or monitor because the males do not respond to the lure chemical, except for a week attraction to cue lures in Western Australian populations. Both sexes may be monitored using protein bait traps (either protein hydrolysate or protein autolysate) (Drew, 1982). Field monitoring is carried out with the utilization of traps within areas of infestation. There is evidence that some fruit flies have different host preferences in different parts of their range; host fruit surveys should be considered as part of the monitoring process.

Ceratitis spp.

Scientific Name (Common Names)
C. capitata (Mediterranean fruit fly)
C. rosa (Natal fruit fly)

Type of Pest
Fruit flies

Taxonomic Position
Class: Insecta, **Order:** Diptera, **Family:** Tephritidae

Reason for inclusion in manual
Pest has a line-item for funding and on Regulated plant pest list

Pest Description
Ceratitis capitata adults are readily recognizable by external morphology, particularly thoracic and wing patterns (White and Elson-Harris, 1994). The males have a characteristically shaped pair of lower orbital setae. The apex is black and diamond-shaped (Fig. 1).

Ceratitis rosa, like other *Ceratitis* spp., has banded wings and a swollen scutellum, which is marked yellow and black (Fig. 1). The pattern of grey flecks in the basal wing cells distinguishes *Ceratitis* spp. from most other genera of tephritids.

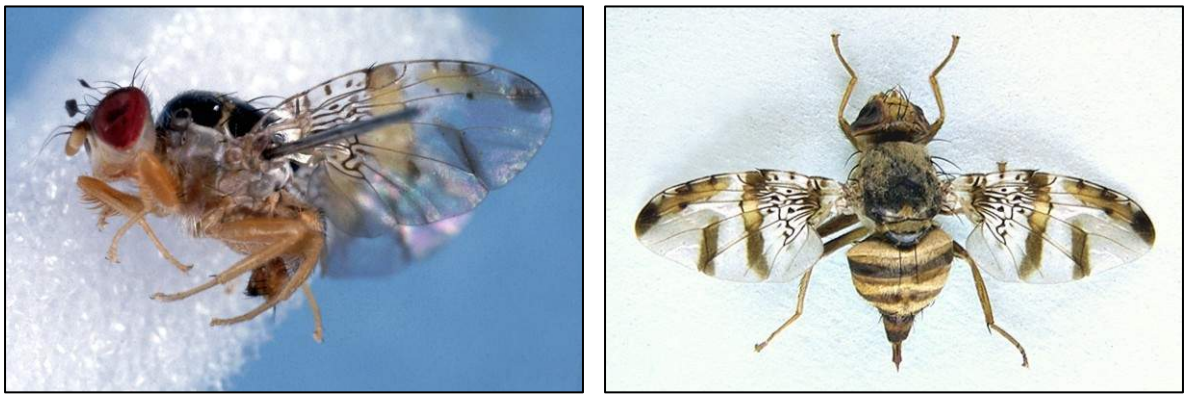


Figure 1. *C. capitata* (left) and *C. rosa* (right). Photos courtesy of CABI, 2004.

Pest Importance
Ceratitis capitata is an important pest in Africa and has spread to almost every continent to become the single most important pest species in its family. It is highly polyphagous and causes damage to a very wide range of unrelated fruit crops. In Mediterranean countries, it is particularly damaging to citrus and peach. It may also transmit fruit-rotting fungi (Cayol et al., 1994).

Damage to fruit crops is frequently high and may reach 100% (Fimiani, 1989; Fischer-Colbrie and Busch-Petersen, 1989). In Central America, losses to coffee crops were

estimated at 5 to 15%; the berries will mature early and fall to the ground with reduced quality (Enkerlin et al., 1989). As in areas where the fly is endemic, in outbreak conditions, the economic impacts include reduced production, increased control costs and a loss of markets.

Ceratitis rosa is highly polyphagous and causes damage to a wide range of unrelated fruit crops. It tends to displace *C. capitata* in some areas where both species occur (Hancock, 1989).

In South Africa, the Natal fruit fly rank second in importance to the Mediterranean fruit fly and, at times, is an even more serious pest. For example, 50 to 100% of plums were reportedly infested in a South African locality one year, despite the control measures applied. The Natal fruit fly has been intercepted in the United States in a shipment of peaches arriving from South Africa; however, it has not been captured as an escapee in the United States. It continues to constitute a potential threat to Florida agriculture. If it were accidentally introduced into Florida and allowed to gain a foothold, the Natal fruit fly could prove to be a serious menace, just as the Mediterranean fruit fly (UFIFAS/FDACS, 2002).

Symptoms

Attacked fruit usually shows signs of oviposition punctures. There is laboratory evidence of fungal transmission. Suspect fruits should be cut open and checked for larvae (Fig. 2). Very sweet fruits may produce a sugary exudate.

Figure 2. *Ceratitis capitata* larvae inside peach fruit (left) and an adult female pumps eggs through her ovipositor into the soft outer layers of a ripe coffee berry (right). Photos courtesy of Florida Division of Plant Industry and S. Bauer, USDA ARS.

Known Hosts (from CABI, 2004)

The Mediterranean (medfly) fruit fly (*C. capitata*) attacks more than 260 different fruits, flowers, vegetables, and nuts. Thin-skinned, ripe, succulent fruits are preferred. Host preferences vary in different regions. Although several species of cucurbits have been recorded as medfly hosts, they are considered to be very poor hosts. Some hosts have been recorded as medfly hosts under laboratory conditions and may not be attacked in the field. Knowledge of the hosts in one country often aids in predicting those that are likely to be infested in a new country, but what may be a preferred host in one part of the world may

be a poor host in another. Primary hosts include bell pepper, *Citrus* spp., coffee, fig, apple, stone fruit, guava, and cocoa.

The Natal fruit fly (*C. rosa*) infests a variety of orchard and wild fruits. Its primary hosts include *Citrus* spp. and Coffea (coffee). Other common hosts include peach, nectarine, apricot, plum, apple, pear, quince, persimmon, fig, guava, avocado, and mango. Nut crops appear to be immune from attack.

Known Distribution (Potential Distribution within the US)

Ceratitis capitata is widespread in Africa and is endemic to most sub-Saharan countries. It has been recorded in Iran, Isreal, Jordan, Lebannon, Saudi Arabia, Syria, Turkey, Yemen, Albania, Croatia, Cyprus, France, Greece, Italy, Malta, Portugal, Spain, Switzerland, most African countries, Costa Rica, El Salvador, Guatamala, Honduras, Nicaragua, Panama, Argentina, Bolivia, Brazil, Columbia, Ecuador, Paraguay, Peru, Uruguay, and Venezuala (Hancock et al., 2001; CABI, 2004). It has been intermittently found in California since 1975, Florida since 1929, and Texas since 1966 (Gasparich et al., 1997).

In 1887*, Ceratitis rosa* was described from specimens collected at Delagoa Bay, Mozambique. By 1900, it was recognized as a pest of orchard fruits throughout much of KwaZulu Natal Province, the Republic of South Africa, and is considered to be the most common fruit fly of economic importance in Rhodesia. Following its accidental introduction into the island of Mauritius about 1953, this fly became firmly established and largely replaced the Mediterranean fruit fly, *Ceratitis capitata*, as a pest of fruits. It has been reported in Angola, Congo, Ethiopia, Guinea, Kenya, Malawi, Mali, Mauritius, Mozambique, Nigeria, Reunion, Rwanda, South Africa, Swaziland, Tanzania, Uganda, Zambia, and Zimbabwe (CABI, 2004).

Survey

A primary method of collecting larvae of *C. capitata* is by cutting infested fruit. Fully grown larvae, when the surrounding air temperature is warm, “flex and jump" repeatedly, as much as 25 mm, when removed from fruit. Adults are primarily collected by use of sticky-board and baited traps. Larval identification is extremely difficult, so that when feasible, it is best to rear them to adults for identification. If collected larvae must be killed, they should be placed in hot water and then transferred to 70% isopropenol. Larval identification is primarily based on characters of mature 3rd instar larvae.

Traps: *Ceratitis capitata* and *C. rosa* can be monitored by traps baited with male lures. As in other tested species belonging to the subgenus *Ceratitis*, males are attracted to trimedlure and terpinyl acetate, but not methyl eugenol. According to IAEA (2003), trimedlure, protein baits (nulure, torula, buminal, *etc*.), and a mixture of ammonium acetate, putrescine, and trimethylamine, can be used as attractants for *C. capitat*a. The same attractants can be used for *C. rosa*, with the exception of the protein baits. These attractants can be used in a Jackson or multilure trap (plastic McPhail type trap).

Trimedlure (t-butyl-4(or 5)-chloro-2-methyl cyclohexane carboxylate) is the most widely used lure for *C. capitata.* The lure is usually placed on a cottonwool wick suspended in the middle of a plastic trap that has small openings at both ends. The lure can either be mixed with an insecticide or a piece of paper can be dipped in dichlorvos and placed in the trap.

Ceralure is also a new potent and persistent attractant for *C. capitata* (CABI, 2004). Traps are usually placed in fruit trees at a height of about 2 m above ground and should be regularly emptied, as it is possible to catch hundreds of flies in a single trap left for just a few days; however, a lure remains effective for a few weeks. Trapping efficiency may also be enhanced by the use of fluorescent colors, particularly light green (CABI, 2004)

Leafhoppers

Homalodisca coagulata

Scientific Name:
Homalodisca coagulata (Say)

Common Name
Glassy-winged Sharpshooter

Type of Pest
Leafhopper

Taxonomic Position
Class: Insecta **Order:** Hemiptera **Family:** Cicadellidae

Reason for inclusion in manual
Emerging Plant Pest, Potential vector for *Xylella fastidiosa*

Pest Description
Eggs: Females lay their eggs in masses of about 10 to 12 on the lower leaf surface of young, fully developed leaves. When it is first laid, the egg mass appears as a greenish blister on the leaf. The female covers the inserted egg mass with a secretion that resembles white chalk (Fig. 1) and is more visible than the leaf blister. Shortly after the eggs hatch, the leaf tissue begins to turn brown. The dead leaf tissue remains as a permanent brown scar.


Figure 1. Egg masses and nymph of *H. coagulata*
Photo courtesy of CABI, 2004.

Nymphs: Nymphs (Fig. 1) look similar to the adults, except they are smaller, wingless, uniform in color (olive-gray), and have prominent bulging eyes.

Adults: Adults (Fig. 2) are about 1.5 to 2.0 cm in length and are generally dark brown to black when viewed from the top or side. The abdomen is whitish or yellow. The head is brown to black and covered with numerous ivory to yellowish spots.

Figure 2. Adults of *H. coagulata.* Courtesy of D. Bartels, USDA APHIS PPQ

Pest Importance

The glassy-winged sharpshooter (GWSS) *H. coagulata* is a xylem feeding leafhopper that is a serious pest because it vectors a strain of the bacterium *Xylella fastidiosa*, the causal agent of Pierce's Disease of grapevines. In Texas, the single greatest threat to the production of susceptible grape cultivars is Pierce's Disease. The disease has caused millions of dollars in losses to the state's wine industry since 1990 and the problem has escalated in the past five years. Within the last few years, the GWSS has established itself in southern California, where it poses a potentially serious threat to the entire wine and table grape industry in the region. Pierce's Disease is caused when *X. fastidiosa* resides, multiplies and interferes with the water-conductive system or xylem of the plant, initially causing dieback of leaves and shoots, and eventually causing the entire plant to collapse and die within one to two years. Strains of this bacterium are vectored by several species of sharpshooters that have also been associated with other diseases, such as plum leaf scald, almond leaf scorch, periwinkle wilt, oleander leaf scorch, ragweed stunt, citrus variegated chlorosis and coffee leaf scorch.

Symptoms

Even though this insect is large enough to be seen with the naked eye, it is very inconspicuous in nature. The brown coloration of the insect blends well with the color of the twigs where it is usually found. This insect can hide by moving to the other side of the twig or branch when it detects movement or is otherwise approached or disturbed. Leaves or fruit can be coated with a whitish, powdery material, which indicates the possibility of heavy GWSS feeding.

Known Hosts

Over 200 host plants in 35 families are known to serve as hosts for *H. coagulata* (CDFA, 1991). Known hosts include avocado, citrus, grape, macadamia, peach, eucalyptus, and many woody ornamentals, such as crape myrtle and sumac.

Known Distribution

Turner and Pollard (1959) describe the range of *H. coagulata* as a strictly southern species native to the US; it is found in abundance from the latitude of Augusta, Georgia to Leesburg, Florida, having a western boundary of Val Verde and Edwards counties in Texas. Current

distribution data shows that the sharpshooter is present in northern Mexico and south Florida. Sorensen and Gill (1996) noted that the range of *H. coagulata* extends to include several counties in southern California, most likely introduced through the nursery industry. The current California distribution includes Kern, Tulare, Ventura, Riverside, San Bernardino and Imperial counties.

Potential Distribution in the US

Homalodisca coagulata has the potential to extend its distribution northward in California to include Solano, Butte, Fresno, and Santa Clara counties.

Survey

Visual surveys can be conducted by examining the terminal leaves, petioles, and stem in citrus and grapes. GWSS infestations can also be determined by examining the underside of plant leaves or recently matured foliage (older foliage should be avoided) for the presence of egg masses. Active stages may be found by searching the plant stems. This stage is easily detected by placing a tarpaulin under the suspected host plant, when temperatures are below 15°C, and striking the plant vigorously. Sweep nets can be used in a similar manner and are used to monitor for glassy-winged sharpshooter in agricultural situations. Yellow sticky traps are commonly used for surveillance and detection of adults in orchards, although *H. coagulata* only shows slight, if any, preference for yellow compared to other colors (CABI, 2004). On warm nights, *H. coagulata* is attracted to black and incandescent lights.

For more detailed survey information, see the California Department of Food and Agriculture:
http://www.cdfa.ca.gov/phpps/pdcp/docs/Survey%20Protocols%202004.pdf

Key Diagnostics

Adults of *H. coagulata* appear dark brown to black in their overall appearance. The abdomen ranges in color from white to yellow and wings have red wing veins. The head is brown to black and covered with numerous ivory to yellowish spots. These spots help distinguish the glassy-winged sharpshooter from a close relative, the smoke-tree sharpshooter *(H. lacerata)*, which is native to the desert region of southern California.

Mealy Bugs

Cataenococcus hispidus

Scientific Name:
Cataenococcus hispidus Morrison

Synonyms:
Erium hispidum, *Ficus retusa, Paraputo hispidus, Planococcus hispidus, Pseudococcus hispidus, Pseudococcus jacobsoni*

Common Name:
Cocoa Mealybug

Type of Pest:
Mealy Bug

Taxonomic position:
Class: Insecta **Order:** Hemiptera **Family:** Pseudococcidae

Reason for inclusion in manual
National pest list

Pest description:
Limited information is available for this mealybug. Mealybugs, in general, are soft-bodied insects that have piercing-sucking mouthparts and that possess a covering of flocculent, white, waxy threads. They have a pink appearance and are covered by powdery 'mealy'-like wax. They can occur on stems and fruit of the host plant. *Cocoa hispidis* is typically attended by the ants *Dolichoderus thoracicus* (the black cocoa ant) (Fig. 1).

Pest Importance
Mealybugs are plant suckers and are serious pests of many agricultural crops. A by-product of mealybug feeding is sticky honeydew, which coats infested foliage and provides an excellent medium for the growth of black sooty mold fungi. This black coating further renders affected plants unsightly and reduces photosynthesis. Ants tend the mealybugs and use the honeydew as a food source; as a result, they scare away predators and parasitoids of the mealybugs.

Figure 1. Black cocoa ant tending *C. hispidus* mealybugs. Droplets of honeydew are food for the ant. Photo courtesy of K.C. Khoo.

On cocoa, *C. hisbidus* feeds by sucking sap from the pod peduncle, pod, and other parts of the tree, but no damage is apparent. Because the black cocoa ants are able to reduce damage caused by other pests of cocoa, this mutualistic relationship between the mealybug and ant is exploited for the biological control of cocoa pests (Ho and Khoo, 1997).

Symptoms

Mealybugs suck plant sap by inserting long piercing-sucking mouthparts deep into plant tissue. They do not cause significant injury to plants in low numbers. Large numbers of mealybugs cause leaf yellowing, leaf curling, and/or leaf drop. Mealybugs are difficult to control because crawlers typically wedge themselves in plant roots, crotches, and leaf folds where pesticides cannot reach them. Plants infested with mealybugs do not always show obvious signs of infestation until it is too advanced to use biological or other control methods.

Small, fluffy, white lumps of about 5mm (1/4") can appear on plants, often in the axil (where the leaf meets the stem), and sticky honeydew and black sooty molds are often present as well.

Known hosts

Annona cherimola (cherimoya), *Citrus spp*., *Nephelium lappaceum* (rambutan)
Theobroma cacao (cocoa), *Saccharum officinarum* (sugar cane)

Known distribution

Malaysia, Thailand

Survey procedures

There are no specific survey methodology established for *C. hispidus*. In the US, surveys for mealybugs require “time-consuming and often laborious examination of plant material for the presence of live mealybugs.” Host tissue should be inspected for small, fluffy, pinkish lumps, the presence of black sooty mold, and tending ants as an indication of mealybug infestation.

Planococcus lilacinus

Scientific Name
Planococcus liliacinus Cock

Synonyms:
Dactylopius coffeae, *D. crotonis, Planococcus crotonis, P. deceptor, P. tayabanus*, *Pseudococcus coffeae, P. crotonis, P. deceptor*, *P. lilacinus*, *P. tayabanus*, *Tylococcus mauritiensis*

Common Name(s)
Oriental mealybug, cacao mealybug, coffee mealybug

Type of Pest:
Mealybug

Taxonomic Position
Class: Insecta **Order:** Hemiptera **Family:** Pseudococcidae

Reason for inclusion in manual
National pest list and Western Region pest list

Pest description
Adult Females: Reliable identification requires detailed study of slide-mounted adult females. In the field, the adult females of *P. lilacinus* may be easily distinguished from *P. citri* by the much more globose shape of *P. lilacinus* (Fig. 1). Beneath the pink to purple wax coating, the body is yellowish. The mid-dorsal line is fairly wide, but indistinct. Illustrations of external features are available in Le Pelley (1968).

Figure. 1. *P. lilacinus* (2.7mm) on cocoa pod. Photo courtesy of CABI, 2004.

The mounted female is broadly oval to rotund, length 1.2 to 3.1 mm, width 0.7 to 3.0 mm. The margin of the body has a complete series of 18 pairs of cerarii, usually all with stout conical setae. Legs stout: hind trochanter + femur 210 to 315 µm long, hind tibia + tarsus 210 to 275 µm long, ratios of lengths of hind tibia + tarsus to hind trochanter + femur 0.77 to 0.97; translucent pores present on hind coxae and tibiae. The inner edges of ostioles are strongly sclerotized. Circulus large, and quadrate, width 105 to 200 µm. Cisanal setae noticeably longer than anal ring setae. Anal lobe cerarii each situated on a moderately sized, well-sclerotized area.

Venter
Multilocular disc pores occur on the median area only and are present around the vulva as single or double rows across posterior borders of median areas of abdominal segments IV to VII and usually in a single row across anterior edge of segment VII (although the latter is sometimes reduced to a few pores) and a few pores sometimes present on anterior edges of median areas of abdominal segments V and VI.

Dorsum
Multilocular disc pores and tubular ducts absent. Setae very long, stout and flagellate (a character which distinguishes it from many *Planococcus* species). Length of longest setae on abdominal segments VI or VII, 50 to 140 µm.

Other Developmental Stages
Descriptions of other developmental stages (eggs, nymphs, male pupae and adult males) are lacking, with the exception of a brief description of the life history by van der Goot (1917).

Pest Importance
Planococcus lilacinus is a foreign plant pest that attacks at least 96 different species of plants, including agricultural and ornamental plants. *Planococcus lilacinus* is a pest of cocoa throughout the Oriental Region and South Pacific area, causing severe damage to young trees by killing the tips of branches. It occurs on and can cause damage to a wide variety of economically important crops, such as coffee, tamarinds, custard apples (*Annona reticulata*), coconuts, Citrus, grapes, guavas and mangoes. It has increased and spread to most coffee-growing areas, attacking the roots and shoots and causing serious damage to the plant. The importance of the species has warranted its control using chemicals and biological control agents in several parts of India, mainly on coffee, cocoa, custard apples and mandarins.

Symptoms
There is very little information on the symptoms of attack by *P. lilacinus* in literature; however, it can cause severe damage and is listed as being a serious pest to coffee, tamarinds, custard apples, coconuts, cocoa and citrus.

Symptoms on coconuts and cocoa are described as button nut shedding, the drying up of inflorescence and the death of tips of branches. Dense colonies form conspicuous patches on fruits; copious honeydew excretion may result in sooty mold development near colonies and the attraction of attendant ants (Watson et al., 1995). Fruits have been reported to have an abnormal shape and to prematurely drop.

Known hosts
Planococcus lilacinus has an extremely wide host range. *Planococcus lilacinus* attacks cocoa, guavas, coffee and other tropical and sub-tropical fruits and shade trees. Its chief hosts, apart from cocoa, appear to be *Annona muricata* (soursop), *Psidium guajava* (guavas), *Citrus spp.*, *Ceiba pentandra* (copal), *Coffea spp.* (coffee), *Coffea arabica* (arabica coffee), *Coffea canephora* (robusta coffee), *Psidium guajava* (common guava),

Tamarindus indica (Indian tamarind), *Vitis spp.,* and species of *Bauhinia*, *Spondias* and *Erythrina* (Le Pelley, 1943).

Cox listed 45 host plant species within 23 families, including Anacardiaceae, Annonaceae, Asteraceae, Bombacaceae, Dioscoreaceae, Dipterocarpaceae, Euphorbiaceae and Fabaceae. Other host plants include bamboos (*Bambusa spp.*) in the Philippines, *Annona squamosa* (sugarapple) in the Philippines and India, and pomegranates, coconuts, tamarinds, *Amaranthus gracilis*, *Ludwigia hyssopifolia*, *Mirabilis jalapa*, *Solanum nigrum*, *Dioscorea* (yam), *Punica granatum* (pomegranate), *Sonchus arvensis*, *Spilanthes acmella*, guavas and mangoes.

Known distribution

Planococcus lilacinus mainly occurs in the Palaearctic, Malaysian, Oriental Australian and Neotropical regions. It is the dominant cocoa mealybug in Sri Lanka and Java. Williams (1982) reports that the species was probably introduced into the South Pacific from Southern Asia. Up to the 1970s, there were no records of it in Africa, but it has been recorded from the Comoros, Kenya and Madagascar. According to Le Pelley (1968), the species does not occur above 1000 m. *Planococcus liliacinus* is also present in Bangladesh, Brunei Darassalam, Cambodia, China, Cocos Islands, India, Indonesia, Japan, Laos, Maldives, Myanamar, Phillipines, Malaysia, Thailand, Vietnam, Yemen, Comoros, Kenya, Madagascar, Mauritius, Dominican Republic, El Salvador, Haiti, Guyana, Papua New Guinea, Guam, Federated states of Micronesia, and the Northern Mariana Islands.

Potential Distribution in the US

Susceptible areas include coastal locations in Mexico, as well as the area abutting the Rio Grande Valley. In the United States, the entire south, extending north and as far as western Pennsylvania, lower Ohio, Indiana, Missouri, and eastern Texas, is susceptible. Even in the colder regions, certain greenhouse crops could be at risk of infestation. For these reasons, Oriental mealybug could become a serious agricultural threat if it were to enter and become established in the United States.

Survey

In the field, *P. lilacinus* may be detected by thoroughly inspecting its normal habitats such as fruits, growing plant tips, shoots and roots. On plants, such as pomegranates, *Annona* and *Citrus*, heavy infestations on fruits can easily be detected (Fig. 2). In quarantine procedures, fruits, plant parts and seedlings of suspect host plants should be thoroughly inspected, if necessary, under a hand lens.

Figure 2. *P. lilacinus* on mango fruit. Photo courtesy of Mango Information network.

Information available on the biology of *P. lilacinus* is very old. It is possible that the species described in Java was a misidentification, as it was barely oviparous because the embryo was fully developed at the time of egg laying. Hatching usually occurs within an hour. The complete life-cycle takes about 40 days in Java. Males pupate on the underside of leaves and are scarce (van der

Goot, 1917).

On cocoa, it is attended by several ant species, including *Dolichoderus bituberculatus* (commonly referred to as the black ant) in Java and *Oecophylla longinoda*, *Technomyrmex detorquens* and *Odontomachus haematodus* in Sri Lanka. In the Philippines it is attended by *Anoplolepis longipes [Anoplolepis gracilipes*], an ant which, on cocoa in Java, tends to displace *D. bituberculatus* and is negatively correlated with *P. lilacinus*.

Planococcus lilacinus has often been mistaken (in the field) for other *Planococcus spp.* on cocoa and coffee, such as *P. citri* and *P. kenyae*, because of its yellowish body color beneath the wax and the presence of a median dorsal stripe extending from the first thoracic to the mid-abdominal region. It can, however, be distinguished by its more globose body shape and by having a much wider, but indistinct, median dorsal stripe.
The slide-mounted female is also quite distinct by the combination of stout legs, long dorsal setae and reduced number of multilocular disc pores that mainly occur in small numbers in the posterior abdominal segments.

Planococcus minor

Scientific Name
Planococcus minor Maskell,

Synonyms:
Planococcus pacificus

Common Name(s)
Passionvine mealybug, Pacific mealybug

Type of Pest
Mealybug

Taxonomic Position
Class: Insecta **Order:** Hemiptera **Family:** Pseudococcidae

Reason for inclusion in manual
National pest list and Eastern Region pest list

Pest Description
This is a small sucking insect with cottony appearance. Females are oval, 1.3 to 3.2 mm in length. The insect body is distinctly segmented and covered with powdery wax, with the appearance of "having been rolled in flour" (Fig. 1).The margin of the body has a complete series of 18 pairs of cerarii, each cerarius with 2 conical setae (except for preocular cerarii which may have 1 or 3 setae). Legs are elongate.

Pest Importance
Pacific mealybug is a foreign plant pest that attacks over 240 different species of plants, including both agricultural and ornamental plants. It has invaded areas in American Samoa, the US Virgin Islands, and Mexico. This pest could enter the eastern United States from Mexico or from the Caribbean, or it could enter California from Mexico or from the Pacific (Vennette and Davis, 2004). The dispersal potential considers both the number of offspring and motility of the pest. On mandarin, this insect completed 10 generations per year and averaged 260 eggs per generation (Sahoo et al., 1999). Local distribution was limited, but over 1,900 interceptions of this pest on various hosts from over 30 countries were reported from 1985 to 2000.

The two polyphagous mealybugs, *Planococcus minor* and *P. citr*i, have similar host ranges and distributions within the Neotropical region and may simultaneously infect the same plant. The predominant species in the South Pacific Islands, the Austro-oriental Region, the Malagasian region, and the Northern Neotropical Region is *P. mino*r, as opposed to *P. citri*, which is present southern states and reported as far north as Ohio, Kansas, and Massachusettes (Cox, 1989). In addition to direct damage, *P. citri* was reported as a virus vector in cocoa, banana, and grape, but whether *P. minor* can serve as a vector is unknown (Jones and Lockhart, 1993; Cabaleiro and Segura, 1997).

Figure 1: Pseudococcidae. Photo courtesy of J. V. French, http://aggiehorticulture.tamu.edu/syllabi/422/pics/arthropd/cm.htm

Symptoms

Mealybugs have piercing-sucking mouthparts. They feed by inserting slender mouthparts into plant tissues and sucking the sap. Plant parts may be spotted, curled, or wilted (Metcalf & Flint, 1939). Infestations reduce the vigor and growth of foliage, which reduces the beauty of the plant and affects marketability (Hamlen, 1975). Mealybugs are a quarantine problem on exported foliage and flowers. This is because species cannot be accurately identified outside the lab, so inspectors/surveyors should treat all specimens as unknown species. There are a large number of endemic species of mealybugs in the US and identifications need to be made by a recognized taxonomic authority.

Known Hosts

Planococcus minor has a broad host range and is considered a non-discriminate feeder within a number of plant families. Citrus (grapefruit, orange, lemon, lime, sour orange), cotton, avocado, guava, mango, cabbage, Chinese cabbage, pineapple, cantaloupe, watermelon, cucumber, pumpkin, soybean, cowpea, beans (kidney, bush, lima), peanut, corn, sugarcane, and grape are hosts of economic importance.

Known Distribution

American Samoa, Australia, Papua New Guinea, French Polynesia, Solomon Islands, Tonga, Western Samoa, Madagascar, Mexico, Bangladesh, Brunei, Indonesia, India, Sri Lanka, Maldives, Philippines, Singapore, Thailand, Taiwan, Vietnam, Argentina, Antigua and Barbuda, Bermuda, Brazil, Colombia, Costa Rica, Cuba, Galapagos islands, Guadeloupe, Grenada, Guatemala, Guyana, Honduras, Haiti, Jamaica, St. Lucia, Suriname, Trinidad and Tobago, Uruguay, and US Virgin Islands.

Potential Distribution within the US

The host range of *P. minor* includes a wide range of plants grown in the United States, so this insect appears capable of establishing populations that mirror the distribution of *P. citri*.

Planococcus citri is present in the southern states and has been reported as far north as Ohio, Kansas, and Massachusettes.

Survey (from Vennette and Davis, 2004)

In the US, surveys for mealybugs other than *P. minor* require "time-consuming and often laborious examination of plant material for the presence of live mealybugs" (Millar et al., 2002). No simple, alternative techniques are available (Millar et al., 2002). The same holds true for *P. minor* surveys in other parts of the world. In India, a regional survey for scales and mealybugs, including *P. minor*, was based on visually examining 25 branches or leaves on each of 15 plants collected from each of 3 field sites in 162 locations (25 x 15 x 3 x 162 = 182,250 leaves examined).

Researchers also depend on visual inspections to assess densities of *P. minor*. In a study of *P. minor* population dynamics, populations of the mealybug were evaluated by visual inspection of citrus leaves, specifically 10 to 15 leaves from 10 randomly selected plants (Bhuiya et al., 2000). Reddy et al. (1997) followed a similar protocol for coffee.

No pheromones have yet been identified for *P. minor*; however, previous research on closely related mealybug species suggests that the identification of a sex pheromone and subsequent development of a pheromone-baited trap is highly feasible.

Key Diagnostics (from Vennette and Davis, 2004)

Planococcus species are not easily distinguishable from one another, especially when immature. A level of complexity is added with variable morphological characters in some species; distinguishing morphological characters can change depending on environmental conditions, such as temperature. *Planococcus citri* and *P. minor* have been taxonomically confused and routinely misidentified as adults because they are similar in appearance and share similar hosts and geographic range (Williams, 1985; Cox, 1989; Williams and Granara de Willink, 1992). Adults (females) can be identified on close examination of morphological characters by a taxonomist. Distinguishable morphological features of closely related mealybug species are described by Cox (1981, 1983, and 1989).

Mites

Eutetranychus orientalis

Scientific Name
Eutetranychus orientalis Klein

Common Name(s)
Citrus brown mite, oriental mite, oriental red mite, oriental spider mite

Type of Pest
Mite

Taxonomic Position
Class: Arachnida, **Order:** Acarina, **Family:** Tetranychidae

Reason for inclusion in manual
National pest list

Pest Description
Eggs: The eggs of *E. orientalis* are oval or circular (Fig. 1)and flattened, coming to a point dorsally but lacking the long dorsal stalk of other spider mites. Newly laid, the eggs are bright and hyaline, but later they take on a yellow, parchment-like color (Smith-Meyer, 1981).

Figure 1. Eggs (left) and adult (right) E. orientalis. Photos courtesy of Pedro Torrent Chocarro.

Larvae: The average size of the larva of *E. orientalis* is 190 x 120 µm. The protonymph is pale-brown to light-green, with legs shorter than the body with an average size of 240 x 140 µm. The deutonymph is pale-brown to light-green with an average size of 300 x 220 µm.

Adults: Adult females are broad, oval and flattened. They vary in color from pale brown to brownish-green to dark green with darker spots within the body. The legs are about as long as the body and yellow-brown (Fig. 1, 2). The average size is 410 x 280 µm.

Adult males are much smaller than the females. They are elongate and triangular in shape with long legs (leg about 1.5 x body length). The body setae are short and cannot be seen with a 10x lens (Dhooria and Butani, 1984; Smith-Meyer, 1981).

Identification requires examination of cleared and mounted female specimens by transmitted light microscopy. Diagnostic descriptions are given by Jeppson et al. (1975) and Smith-Meyer (1987).

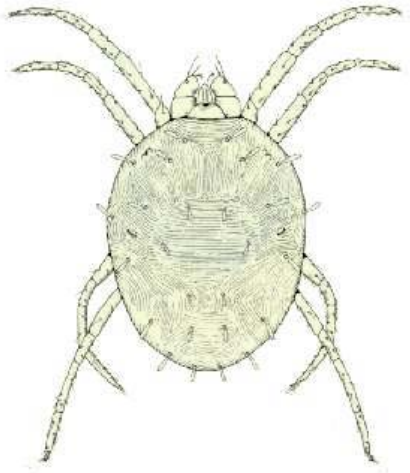


Figure 2. *Eutetranychus orientalis.* Drawing courtesy of CSIRO Entomology, Australia.

Eutetranychus orientalis has the following combination of characters: striae on the prodorsum longitudinal and tuberculated; striae between the second (d/sub/1) and third (e/sub/1) dorsocentral setae longitudinal or V-shaped; the 13 pairs of dorsal body setae all arise from basal tubercles and vary in length and shape; dorsolateral setae on the body (c2), (d2), (e2), (f2) are long, lanceolate and subspatulate or broadly spatulate; dorsocentral setae (c1), (d1), (e1), (f1), (h1) short and spatulate or lanceolate or subspatulate; first pair of dorsocentral setae (c1), first pair of dorsal lateral setae (c2) and humeral setae (c3) all more or less in line; third (e1) and fourth (f1) dorsocentral setae form a square; terminal sensillum (spinneret) of palptarsus three times as long as broad; coxa II with one seta; tactile setal formulae (I-IV): femora 8-6-(3-4)-(1-2), genua 5-5-2-2, tibiae 9-6-6-7; chromosome number (n)=3.

Pest Importance

Eutetranychus orientalis is generally regarded as an important pest of citrus. In India, of the seven species reported as pests on citrus, only *E. orientalis* was reported to be a major pest in all areas (Dhooria and Butani, 1984).

Symptoms

Eutetranychus orientalis begins feeding on the upper side of the leaf along the midrib and then spreads to the lateral veins, causing the leaves to become chlorotic. Pale yellow streaks develop along the midrib and veins. Little webbing is produced. In heavier infestations, the mites feed and oviposit over the whole upper surface of the leaf. Very heavy infestations on citrus cause leaf fall and die-back of branches, which may result in defoliated trees. Lower populations in dry areas can produce the same effect.

Known Hosts

The primary host of *E. orientalis* is *Citrus* spp. Other hosts include peaches (*Prunus persica*), pears (Pyrus), Plumeria, quinces (*Cydonia oblonga*), *Ricinus communis*, sunflowers (*Helianthus annuus*), sweet potatoes (*Ipomoea batatas*), water hyacinth (*Eichhornia crassipes*), watermelons (*Citrullus lanatus*) and over 50 other plant species. In China, *E. orientalis* attacks *Alstonia glaucescens*.

Primary hosts
Abelmoschus esculentus (okra), *Carica papaya* (papaw), *Citrus*, *Codiaeum variegatum* (croton), *Ficus carica* (common fig), *Gossypium* (cotton), *Morus alba* (mora), *Nephelium lappaceum* (rambutan), *Plumeria* (frangipani), *Ricinus communis* (castor bean), and *Solanum melongena* (aubergine).

Secondary hosts
Manihot esculenta (cassava), *Musa x paradisiaca* (plantain), *Olea europaea subsp. europaea* (olive), *Prunus dulcis* (almond), and *Psidium guajava* (common guava).

Known Distribution
Afghanistan, Bangladesh, China, India, Iran, Israel, Japan, Jordan, Lebanon, Pakistan, Philippines, Thailand, Turkey, Yemen, Cyprus, Spain, Cape Verde, Egypt, Kenya, Malawi, Mauritania, Mozambique, Nigeria, Senegal, South Africa, Sudan, Swaziland, and Australia (CABI, 2004).

Potential Distribution within the US
No information available at this time.

Survey
The presence of *E. orientalis* can be detected by discoloration of the host leaves and pale-yellow streaks along the midribs and veins. Adult females are larger than the males. They are oval, flattened and often pale brown through brownish-green to dark green in color.

Moths

Epiphyas postvittana

Scientific Name
Epiphyas postvittana Walker

Synonyms:
Tortrix postvittana, Austrotortrix postvittana, Cacoecia postvittana, Teras postvittana, Archips postvittanus

Common Name(s)
Light brown apple moth, apple leafroller, Australian leafroller

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order**: Lepidoptera, **Family**: Tortricidae

Reason for inclusion in manual
National pest list and Western Region pest list

Pest Description
Light brown apple moth adults are sexually dimorphic and variable in wing pattern and color. A light, diamond-shaped area is visible and extends from behind the head to approximately one-third of body length when the moth is at rest. Male forewing lengths range from 6 to 10 mm, compared with 7 to 13 mm in females (Thomas, 1975). Males tend to have a higher contrast in coloration than females, although the level of contrast varies (Fig 1).

The first instar larvae are approximately 1.6 mm long, and final instar larvae range from 10 to 20 mm in length. The body of a mature larva is green with a darker green central stripe and two side stripes. The first larval instar has a dark-brown head; all other instars have a light-fawn head and prothoracic plate. Overwintering larvae are typically darker (Fig. 2).

Pupae are green after pupation, but become brown within one day.

Figure 1. *Epiphyas postvittana*; adult male (A) and adult female (B). (museum set specimen). Photo courtesy of CABI, 2004.

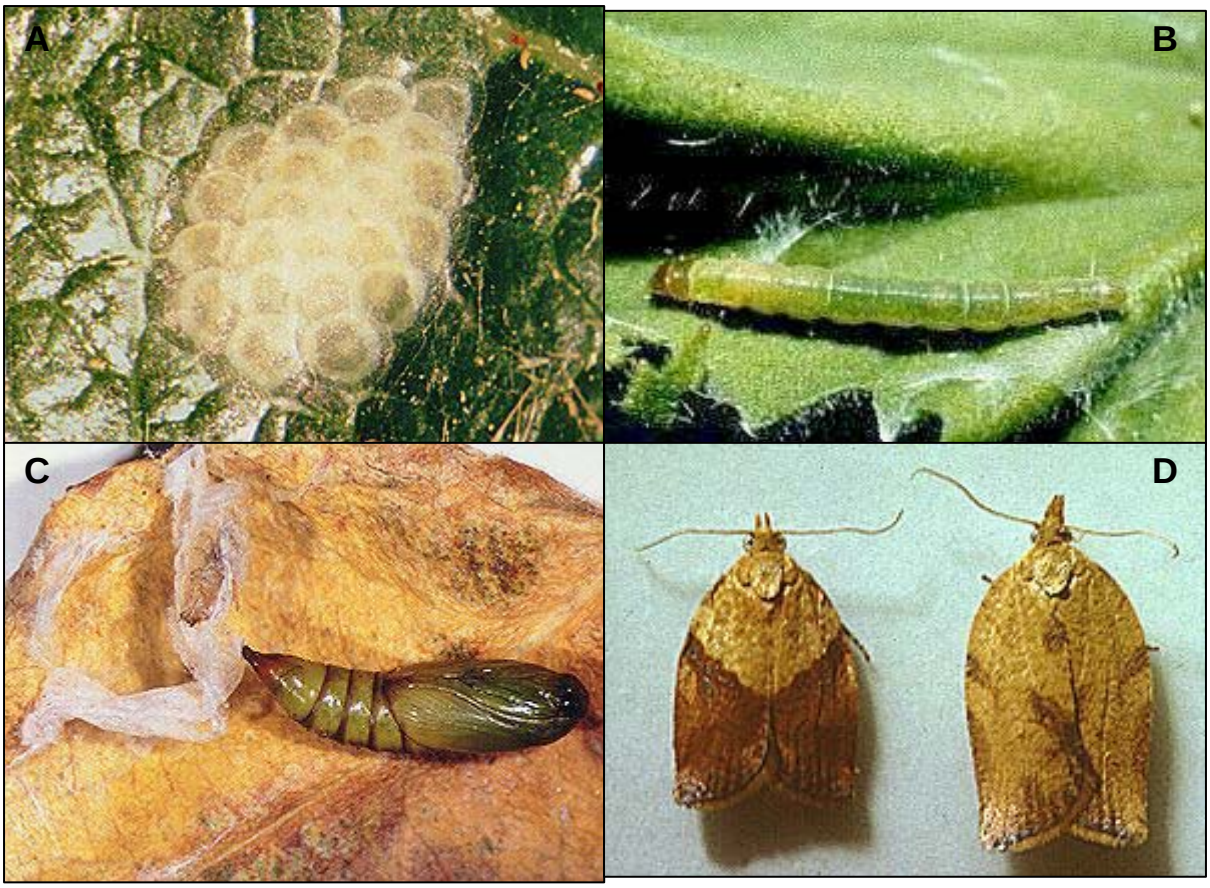


Figure 2. Life stages of *Epiphyas postvittana*: (A) eggs; (B) larva; (C) pupa, (D) adults, male is on the left. (Photos from http://www.hortnet.co.nz/key/keys/info/lifecycl/lba-desc.htm)

Pest Importance

Epiphyas postvittana is a highly polyphagous pest that attacks a wide number of fruits, ornamentals, and other plants. According to Geier and Briese (1981), “Economic damage

results from feeding by caterpillars, which may destroy, stunt or deform young seedlings, spoil the appearance of ornamental plants, and/or injure deciduous fruit-tree crops, citrus, and grapes." Losses in Australia were estimated to be AU$21M per year, but there has been no similar estimation in other countries.

The larvae can be damaging to grape, apple, and peach. In grape, 70,000 larvae/ha were documented to cause a loss of 4.7 tons of chardonnay fruit in 1992 with an estimated cost of $2000/ha (Bailey et al., 1995). A single larva can destroy about 30 grams of mature grapes. Damage to apples is in the form of either pinpricks (flask-shaped holes about 3 mm deep into the fruit) or entries, (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; the second generation damages fruit harvested later in the season. Some varieties of apples include 'Sturmer Pippin' (an early variety), 'Granny Smith' and 'Fuji' (late varieties); they can have up to 20% damage while severe attacks can damage up to 75% of a crop (USDA, 1984). Peaches are damaged by feeding that occurs on the shoots and fruit (Lo et al., 1995).

Mature larvae are the most difficult stage to control (Lay-Yee et al. 1997). *Epiphyas postvittana* is difficult to control with sprays because of its leaf-rolling ability. There is evidence of insecticide resistance due to the overuse of sprays (Geier and Briese, 1981).

Canada has listed *E. postvittana* as a noxious pest. 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 meter-row of fruit crops; however, if the crop is intended for export, control is recommended even if only one larva is found (Charles et al., 1987).

Symptoms

Larval nests are typically seen as leaves webbed together or to fruit. Fruit surface feeding is common within larval nest sites. On apples, older skin damage has a cork-like appearance, and may be small (5 mm) or larger areas, depending on larval instar and feeding duration. Feeding sites on other fruits are similar. Vectoring of the fungus *Botrytis cinerea* by larvae has been shown in grapes causing up to 13% of berry damage (by weight) (Bailey, 1997).

Known Hosts

Epiphyas postvittana has a wide host range, with 73 listed from Australia (Danthanarayana, 1975; Geier and Briese, 1981), and a larger number from New Zealand (Thomas, 1989; Dugdale and Crosby, 1995). Danthanarayana et al. (1995) have suggested that the better performance of *E. postvittana* on herbaceous rather than woody plants suggests that it primarily evolved as a feeder on the former.

In Australia, capeweed, curled dock, and plantain are important hosts. In New Zealand, important perennial weed hosts include gorse (*Ulex europeus*) and broom (*Cytisus scoparius*), although in several regions it has been commonly recorded on annual weeds (*Rumex obtusfolius* and *Plantago* spp.) and on shelter and amenity trees (species of Salix and Populus) (Suckling et al., 1998).

Primary hosts
Actinidia chinensis (Chinese gooseberry), *Chrysanthemum x morifolium*, *Citrus spp.*, *Cotoneaster*, *Crataegus* (hawthorns), *Diospyros* (malabar ebony), *Eucalyptus*, *Feijoa sellowiana* (Feijoa fruit), *Humulus lupulus* (hop), *Jasminum* (jasmine), *Ligustrum vulgare* (privet), *Litchi chinensis* (leechee), *Malus pumila* (apple), *Medicago sativa* (lucerne), *Persea americana* (avocado), *Pinus* (pines), *Pinus radiata* (radiata pine), *Populus* (poplars), *Prunus armeniaca* (apricot), *Prunus persica* (peach), *Pyrus* (pears), *Ribes* (currants), *Rosa* (roses), *Rubus* (blackberry, raspberry), *Solanum tuberosum* (potato), *Trifolium* (clovers), *Vaccinium* (blueberries), *Vicia faba* (broad bean), and *Vitis vinifera* (grapevine).

Known Distribution
Epiphyas postvittana is widespread throughout New Zealand and Australia on many weedy hosts (Suckling et al., 1998). It is commonly present in gardens and unsprayed horticultural crops. In the past, it has been reported in England, Wales and Hawaii (CABI, 2004).

Potential Distribution within the US
No occurrences of *E. postvittana* have been reported in the wild continental US; however, this species has a broad host range and is likely to find suitable climatic conditions in much of the US. It has been reported as present in Hawaii (CABI, 2004).

Survey (Taken from Venette et al, 2003 and CABI, 2004)
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, 1977). 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). 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).

Pheromone traps have been widely used for detection and monitoring populations of this species since the identification of the sex pheromone (Bellas et al., 1983). 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. A range of applications were reported by Suckling (1993), including insecticide resistance monitoring, spray reduction, and sample collection for population studies. Pheromone traps have been proposed for use in the biosecurity detection of this species in the US (Schwalbe and Maestro, 1988). Bradley et al. (1998) reported the use of traps with a spray threshold. Shoot tip assessment has been used on apples and other crops. Suckling et al. (1998) used time searches for alternative host plants.

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 6.5 ft (2 m) 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 5 ft (1.5 m) above the ground, and lures were changed every 6 weeks (Thomas and Shaw, 1982; Suckling et al., 1990; Suckling and Shaw, 1992).

Adults are also attracted to fruit fermentation products. 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) 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).

Key Diagnostics

Epiphyas postvittana is similar to *E. pulla* (Turner) and *E. liadelpha* (Meyr.). Geier and Springett (1976) reported possible hybridization based on demographic characteristics. Larvae are similar to larvae of other leafrollers that may be present (for example, in New Zealand *Planotortrix octo*, *P. excessana, Ctenopseustis obliquana* and *C. herana*). 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).

Eudocima fullonia

Scientific Name
Eudocima fullonia Clerck

Synonyms:
Othreis fullonia

Common Name(s)
Fruit piercing Moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for inclusion in manual
National pest list

Pest Description
The main species active on citrus are *Eudocima fullonia, E. materna* and *E. salaminia*. They have 7 to 10 cm wingspans, with forewings of mottled brown, grey or green and silvery white. The forewings of males and females differ in the first two species. The hind wings are characterized by the orange-yellow color, extensively bordered by a black and hatched area and a central black mark (kidney shaped or round) (Fig. 1). These are often exposed when the moth is feeding. An individual moth can spend several hours feeding from the one fruit, but would generally attack a number on a single night.

Figure 1. Adult female fruit piercing moth. Photo courtesy of CABI, 2004.

Eggs: are hemispherical, just over 1 mm in diameter, and greenish-white to creamy-yellow when laid. Delicate surface sculpturing can be seen with the aid of a microscope. The brownish head capsule of the developing larva becomes obvious beneath the shell a few hours before hatching. Eggs are generally laid on the undersides of leaves of host plants. Most occur singly, but batches of up to several hundred can be laid (in some localities) when moth populations are large (Waterhouse and Norris, 1987).

Figure 2. Larvae of *E. fullonia*. Photo courtesy of CABI, 2004.

Larvae: The newly hatched larvae are 4 to 5 mm long, bright translucent green in color, and inconspicuously banded by brown spots and hairs (Hargreaves, 1936). The head capsule is 0.5 mm wide. Second instars are a uniform dull black, with two developing, paired, lateral orange eyespots. Larvae molt four or five times during development. Final instars can reach about 60 mm in length, with a head capsule of 4.5 mm. Mature larvae are a velvety brown to black or pale yellow to green. There are fine powdery white spots along the entire length of the body and two conspicuous, paired, lateral eyespots on the second and third abdominal segments. In dark larvae, the eyespots are peripherally white (above) and orange (below), with a central black area surrounding a pale blue core. Larvae have four abdominal pairs of prolegs, the initial pair being rudimentary. The anal segment is dilated and bears a tubercle. When resting, larvae hold the posterior part of the body upwards, while the anterior part is curled with the head tucked under (Fig. 2). If disturbed, larvae may rear and 'spit' digestive juices. Larvae move with a semi-looping action.

Pupae: The post-feeding larva forms a silken cocoon among the leaves of the larval host plant and attaches itself within it at the anal end. The pupa is about 30 mm long, a glistening brown-black, and can be sexed at this stage using differences in the position of the genital grooves (Common, 1990).

Adults: Moths are robust, with a wing span of 80 to 100 mm, and are about 3.5 to 5.0 cm long. The thorax is a purplish-brown and the abdomen orange-yellow. The hindwings have an orange-yellow base color and contain a large black, kidney-shaped mark (Fig. 1). The distal margins of the hindwings have a broad, black border, edged with white flecks. The color patterns of the forewings are sexually dimorphic. Males have leaf-like forewings of red-brown to purplish-brown. There is an inconspicuous, irregular spot centrally placed near the anterior margin. In females, the forewings are more variegated and striated than in males. The color varies between purplish-brown and greyish-ochre, often flecked with green and white. There is a distinct dark, roughly triangular mark in a similar position to the spot in the male forewing. Adults rest with the forewings held tent-like over the body. When feeding, the forewings are held out exposing the bright hindwings.

Biology and Ecology:
The duration of the life-cycle from egg to egg-laying adult female is 35 to 49 days in a study conducted in Fiji at temperatures between 82.4 to 91.4 °F (28 to 33° C) and 30 to 33 days in the warmer conditions of New Caledonia (Waterhouse and Norris, 1987). Generations are continuous throughout the year.

The small hemispherical eggs are about 1/25 inch (1 mm) in diameter and are colored yellowish green. When moth populations are low, a single female moth lays her eggs in batches of up to 100. When populations are high, eggs are laid in batches of several hundred eggs by individual females. Eggs are generally laid on the underside of leaves, but may be found on the bark or on other plants nearby. Eggs hatch in 3 to 4 days at temperatures between 82.4 to 91.4 °F (28 to 33 °C).

The female lays up to 300 eggs at a time on the underside of the leaves of the leguminous tree *Erythrina*. The larva molts four times. Pupae are formed among the webbed leaves of *Erythrina* or on nearby plants. They are about 3 cm long and shining brown-black. The life-

cycle from hatching of the egg to adult takes about 30 to 62 days, depending on the season and on other environmental factors.

Caterpillars mostly feed between 5:00 PM and 10:00 AM, but may feed at any time. They are usually located beneath or on the edges of leaves (Hargreaves, 1936). Young larvae drop to the ground at any sign of danger, while the older larvae take an aggressive attitude by hanging on to the food plant with their hind legs and swaying the rest of their body from side to side (Mosse-Robinson, 1968).

Although their flight is slow and heavy (Baptist, 1944), the adult moths are very strong fliers and can travel great distances from their breeding grounds in search of food (Waterhouse and Norris, 1987). Adults mainly fly between the hours of dusk and 11:00 PM. They are not usually attracted to light. When disturbed, the moth flares its forewings, exposing its conspicuous hindwings (Waterhouse and Norris, 1987). This insect is principally a pest in the adult stage. It feeds primarily at night on a wide variety of ripening fruit by piercing the fruit and sucking out the juices. Moth populations are generally higher during the rainy season. Large populations of moths periodically occur following droughts.

Pest Importance

In those countries in which more than one species of *Eudocima* exist, it is difficult to stipulate the proportion of fruit lost to *E. fullonia*. However, in Thailand, it was reported that *E. fullonia* caused an estimated 70 to 90% and 50 to 70% of primary damage inflicted by fruit-piercing moths on longan and citrus, respectively. In northeast Australia, an average 88.8% of moths inflicting damage on lychees and carambolas were *E. fullonia* (Fay and Halfpapp, 1993a). In drier tropical areas, *Eudocima materna* seems to increase in prominence. Although losses of fruit to *E. fullonia* in New Caledonia are minor in regular years, 95% of citrus and 100% of tomatoes have been damaged in outbreak years (Waterhouse and Norris, 1987). In Fiji, 10 to 15% of ripe fruit (primarily citrus) was regularly lost to *E. fullonia*, rising as high as 73% (Waterhouse and Norris, 1987). Irregular outbreaks of *E. fullonia* have threatened entire crops of navel oranges in southeast Queensland (Fay, 1997). Similarly, surveys in parts of China have found 40 to 60% of citrus fruits damaged by *E. fullina*.

Failure to detect fruit-piercing moth damage at harvest or packing can result in sound fruit being contaminated by fermenting juices during shipment. Whole boxes or cartons of fruit may then be lost. The economic impact of fruit-piercing moths is often masked by the emphasis placed on fruit flies, some of which occasionally utilize fruit-piercing moth wounds for oviposition.

Figure 3. Male (left) and female (right) *E. fullonia* on a green mandarin fruit. Photo courtesy of CABI, 2004.

Symptoms

For most moth and butterfly pests, the caterpillars are the damaging stage. The Pacific fruit-piercing moth differs in this aspect, because it is the adult moth that is

the damaging stage, and the larvae are essentially not harmful. The mouth parts of the moth are about an inch (2.5 cm) long and strong enough to penetrate through tough-skinned fruit. Once the moth has punctured the skin of the fruit, a process that usually takes a few seconds, it feeds on the juices of the fruit (Fig. 3). Feeding occurs at night and the fruit does not have to be ripe to be fed upon by this moth (Waterhouse and Norris, 1987). Fruit flesh damaged by this moth becomes soft and mushy differing from fruit damaged by fruit flies, which is more liquid.

Damage caused by this pest is not only a result of the direct feeding of this moth, but also by the fungal and bacterial infections that develop at the wound site. This moth is a known vector of *Oospora citri*, a fungus that rots the fruit and has a penetrating odor that attracts the moth. Other microorganisms that gain entrance into the fruit and cause rotting include *Fusarium* spp., *Colletotrichum* spp., and several types of bacteria. When moths are abundant, the green fruit is attacked, causing premature ripening and fruit drop. On oranges, a green fruit turns yellow at the site of the piercing and fungi soon develops within the wound (Waterhouse and Norris, 1987).

Incidence of damage by this moth is normally low; however, when outbreaks occur, most of the crop is affected. Only occasionally is damage appreciable. The adult moths pierce ripening fruit to obtain sap. The damage looks superficially similar to fruit fly damage, but eggs or fruit fly larvae cannot be found. Fruits are generally pierced as they mature and ripen, but green fruit is also susceptible if moth populations are high and ripe fruit is limited. A round, pinhole-sized puncture is made in fruits. The hole serves as an entry point for disease organisms and can result in early fruit drop. The latter is an obvious sign of fruit piercing moth activity in citrus. A small cavity is left in the fruit in the feeding site. The area of the fruit around the cavity will be dry and spongy.

Damage is difficult to immediately detect after it occurs. Close inspection of a fruit will reveal a neat pin-sized hole, which, when pressed, will usually squirt juice. A sizeable area beneath the skin is bruised and often honeycombed. Fruit rot organisms rapidly invade the wound causing fermentation and breakdown. In mature fruit, a large, round, brown spot develops on the skin around the piercing site. Damaged fruits are completely unmarketable and must be removed at packing to avoid contamination of sound products.

Known Hosts

The Pacific fruit-piercing moth attacks many fruit and vegetable crops. Fruit crops attacked include apples, apricots, bananas, breadfruit, coffee, figs, grapefruit, grapes, guava, kiwifruit, lemon, lime, litchi, longan, mandarins, mangoes, nectarines, oranges (especially navels), papaya, passion fruit, peaches, persimmons, pineapple, plums, pummelo, and star fruit. Vegetable crops attacked include eggplant, tomatoes, pepper, and melons (Waterhouse and Norris, 1987).

Except in the Pacific, larvae develop on plants belonging to the Menispermaceae family, especially the creepers belonging to the *Tinospora*, *Tiliacora*, *Triclisia*, and the *Stephania* genuses. In the Pacific, the larvae of this moth almost exclusively develop on plants belonging to the *Erythrina* genus, or false wiliwilis, a common landscaping plant. The only exception is the creeper, *Stephania forsteri* (Menispermaceae). In Hawaii, larvae have been found on *Erythrina fusca* and *E. variegata orientalis*.

Known Distribution

This moth is native to the Indo-Malaysian region, and widespread throughout the Pacific basin, Asia, and Africa. It is not recorded in the Americas. Some countries where it is found include American Somoa, Angola, Australia, Cambodai, Cameroon, China, Congo, Fiji, Ghana, Guam, India, Indonesia, Japan, Kenya, Korea, Laos, Liberia, Madagascar, Malawi, Mongolia, Monzambique, Nemibia, Nepal, Nigeria, Pakistan, Papua New Guinea, the Philippines, Reunion, Samoa, Sierra Leone, Singapore, Sri Lanka, Tanzania, Thailand, Tonga, Uganda, Vietnam, and Zimbabwe. Refer to Waterhouse and Norris (1987) for an extensive list of countries where this moth is present. It was first reported in Hawaii on Oahu in 1985, and by 1986, it was present on Kauai, Hawaii, Maui and Molokai.

Potential Distribution within the US

At this time, the moth is absent in the US, with the exception of Hawaii.

Survey

The most effective way to monitor for fruit-piercing moths is to inspect the crop by torchlight after sundown a few weeks before harvest. The large, red-glowing eyes of the moths are easily seen. Large traps, such as the Gamgee trap (Vock, 1990), are sometimes used in detection and control, but their efficiency is questionable at a commercial level. Up to 20 traps are used per hectare, which are placed along the orchard's edge. Ripe to over-ripe bananas are normally used to the bait traps.

In some fruits, such as lychees, detection of fruit-piercing moth damage can be difficult. The slightest sign of weeping can be an indication; with this, when the fruit is pressed it will usually squirt out juice. The damage site will be flaccid and flattened in appearance, and lack the firm, rounded flesh of intact fruit. Many farmers opt to place freshly picked fruit in cool storage at high humidity, which facilitates the detection of damage after one day. Feeding attractant studies have shown that some species, such as *E. fullonia,* are most responsive to the general fruity esters fruits produce. These esters are very volatile and contribute an increasing proportion to fruit odor as ripening occurs. Other volatile compounds, particularly some aldehydes and alcohols, are important to the attraction process by contributing to the 'greenness,' which determines the level of ripeness. In fact, tests have shown that moths seek out fruits of a particular stage of maturity based solely on proportions of the volatile components produced.

Combinations of attractants have been developed and incorporated into sugared-agar baits. Field trials with a range of these experimental baits in Clementine and hybrid mandarins in north Queensland showed many more attacks on the best baits (85% of moths) than on fruit up to the early harvest phase. Baits were less competitive, but still very attractive, when nearly all fruit reached the ripe stage. Recent studies have resulted in the selection of a suitable toxicant to incorporate in the baits, while the means to present the attractants so that they have a practical field life are still being pursued. Tests are also being conducted in crops other than citrus. The technique will then move into a commercial development phase.

Key Diagnostics

Adults of *E. fullonia* closely resemble species such as *Eudocima homaena* and *E. jordani* (Moore, 1881). All species of *Eudocima* cause similar damage. Separation of species involves detailed microscopic examination. Fruit discoloration could be attributed to fruit fly damage, but the size of the hole left at the damage site will clarify whether fruit-piercing moths were involved.

Helicoverpa armigera

Scientific Name
Helicoverpa armigera Hübner

Common Name(s)
Old world bollworm, scarce bordered straw worm, corn earworm, African cotton bollworm. American bollworm, tomato worm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for inclusion in manual
National pest list and Eastern Region pest list

Pest Description
Egg: Yellowish-white and glistening at first (Fig. 1A), changing to dark-brown before hatching; pomegranate-shaped, 0.4 to 0.6 mm in diameter; the apical area surrounding the micropyle is smooth, the rest of the surface sculptured in the form of approximately 24 longitudinal ribs, alternate ones being slightly shorter, with numerous finer transverse ridges between them; laid on plants which are flowering, or are about to produce flowers.

Larva: The first and second larval instars are generally yellowish-white to reddish-brown in color, without prominent markings; the head, prothoracic shield, supra-anal shield and prothoracic legs are very dark-brown to black, as are also the spiracles and tuberculate bases to the setae, which give the larva a spotted appearance (Fig. 1B); prolegs are present on the third to sixth, and tenth, abdominal segments. A characteristic pattern develops in subsequent instars. Fully grown larvae are approximately 30 to 40 mm long; the head is brown and mottled; the prothoracic and supra-anal plates and legs are pale-brown, and only the claws and spiracles are black; the skin surface consists of close-set, minute tubercles. Crochets on the prolegs are arranged in an arc. The final body segment is elongated. Color pattern: a narrow, dark, median dorsal band; on each side, first a broad pale band, then a broad dark band; on the lateral line, a broad, very light band on which the row of spiracles shows up clearly. The underside is uniformly pale. On the basic dorsal pattern, numerous very narrow, somewhat wavy or wrinkled longitudinal stripes are superimposed. Color is extremely variable and the pattern described may be formed from shades of green, straw-yellow, and pinkish- to reddish-brown or black.

Pupa: Mahogany-brown, 14 to 18 mm long, with a smooth surface, rounded both anteriorly and posteriorly, with two tapering parallel spines at posterior tip.

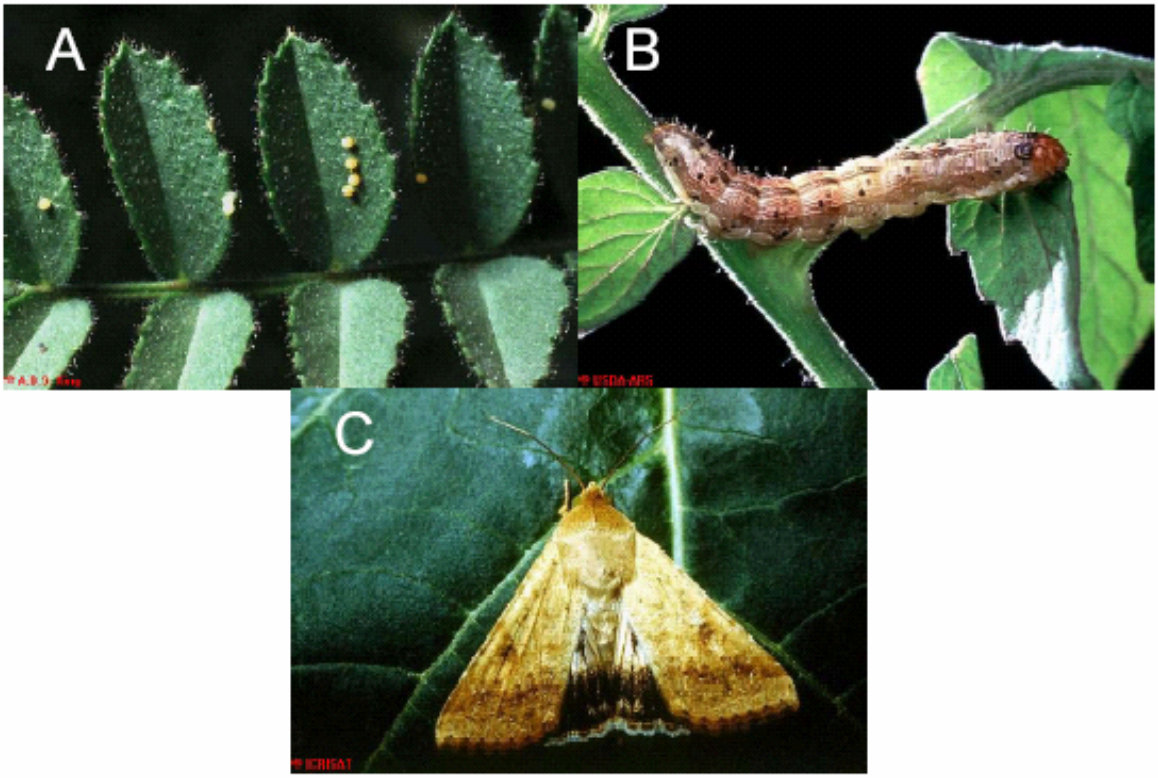


Figure 1. Life stages of *Helicoverpa armigera*, images not to scale: (A) eggs; (B) larva; and (C) adult. Photos courtesy of CABI, 2004.

Adult: Stout-bodied moth of typical noctuid appearance (Fig. 1C) with a 3.5 to 4 cm wing span; broad across the thorax and then tapering, 14 to 18 mm long; their color is variable, but males are usually greenish-grey and female orange-brown. Forewings have a line of seven to eight blackish spots on the margin and a broad, irregular, transverse brown band. Hindwings are pale-straw colored with a broad dark-brown border that contains a paler patch; yellowish margins with strongly marked veins and a dark, comma-shaped marking in the middle are evident. Antennae are covered with fine hairs.

(For more information on descriptions see Dominguez Garcia-Tejero (1957), Hardwick (1965), Cayrol (1972), Delatte (1973), King (1994).)

Pest Importance

Helicoverpa armigera, like its close relatives *H. zea* and *Heliothis virescens* in the New World, is a pest of major importance in most areas where it occurs, damaging a wide variety of food, fibre, oilseed, fodder and horticultural crops. Its considerable pest significance is based on the peculiarities of its biology, mobility, polyphagy, and its rapid and high reproductive rate. Diapause makes it particularly well-adapted to exploit transient habitats, such as man-made ecosystems. Its predilection for harvestable flowering parts of high-value crops, including cotton, tomato, sweet corn and the pulses, confers a high economic

and socio-economic cost in subsistence agriculture due to its depredations. Regional and even relatively local differences in host preference can give rise to differences in pest status on particular crops; this was shown by populations in northern and southern India where severe infestations of cotton are only a recent event (CABI, 2004).

Symptoms

On Cotton: Bore holes are visible at the base of flower buds, the latter being hollowed out. Bracteoles are spread out and curled downwards. Leaves and shoots may also be consumed by larvae. Larger larvae bore into maturing green bolls; young bolls fall after larval damage. Adults lay fewer eggs on smooth-leaved varieties.

On Tomatoes: Young fruits are invaded and fall; larger larvae may bore into older fruits. Secondary infections by other organisms lead to rotting.

On Maize: Eggs are laid on the silks, larvae invade the cobs and developing grain is consumed. Secondary bacterial infections are common.

On Sorghum: Larvae feed on the developing grain, hiding inside the head during the daytime. Compact-headed varieties are preferred.

On Chickpea: Foliage, sometimes entire small plants consumed; larger larvae bore into pods and consume developing seed. Resistant cultivars exist.

On Pigeon pea: Flower buds and flowers bored by small larvae may drop; larger larvae bore into locules of pods and consume developing seed. Short duration and determinate varieties are subject to greater damage. Less-preferred varieties exist.

On Groundnut: Leaves, and sometimes flowers, are attacked by larvae; severe infestations cause defoliation. Less preferred varieties exist (CAB, 2004).

Known Hosts

Primary hosts

Abelmoschus esculentus (okra), *Allium* (onions, garlic, leek, etc.), *Arachis hypogaea* (groundnut), *Avena sativa* (oats), *Brassicaceae* (cruciferous crops), *Cajanus cajan* (pigeon pea), *Capsicum annuum* (bell pepper), *Cicer arietinum* (chickpea), *Citrus*, *Cucurbitaceae* (cucurbits), *Glycine max* (soybean), *Gossypium* (cotton), *Helianthus annuus* (sunflower), *Hordeum vulgare* (barley), *Lablab purpureus* (hyacinth bean), *Linum usitatissimum* (flax), *Lycopersicon esculentum* (tomato), *Mangifera indica* (mango), *Nicotiana tabacum* (tobacco), *Pennisetum glaucum* (pearl millet), *Phaseolus* (beans), *Phaseolus vulgaris* (common bean), *Pinus* (pines), *Pisum sativum* (pea), *Polyphagous* (polyphagous), *Prunus* (stone fruit), *Solanum melongena* (aubergine), *Solanum tuberosum* (potato), *Sorghum bicolor* (common sorghum), *Triticum* (wheat), *Triticum aestivum* (wheat), *Vigna unguiculata* (cowpea), and *Zea mays* (maize) (CAB, 2004).

Wild hosts

Acalypha (Copperleaf), *Amaranthus* (grain amaranth), *Datura*, *Datura metel* (Hindu datura), *Gomphrena*, and *Hyoscyamus niger* (black henbane) (CAB, 2004).

Known Distribution

Helicoverpa armigera is found in the Palearctic, Oriental, Ethiopian, and Austalian zoogeographis provinces, south of a line at approximately 52°N. This range occupied by the species includes tropical, dry, and temperate climates (CABI, 2004). The currently reported global distribution of *H. armigera* suggests that the pest 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 forest.

Potential Distribution within the US

Based on the distribution of climate zones in the US, approximately 49% of the continental US would be suitable for *H. armigera*. Despite the number of *H. armigera* that are introduced into the US each year, no occurrences of the pest have been reported in the wild. A wide variety of factors may contribute to the failed establishment of any introduced population, thus. it is generally recognized that biological invasion is a difficult, unlikely event. Nevertheless, we must acknowledge the other possibility, that *H. armigera* has in fact already established (conceivably small, non-damaging) populations that have gone unnoticed or been misidentified as another *Helicoverpa*/*Heliothis* species (CABI, 2004).

Survey

This polyphagous moth is one of the principal enemies of cotton and maize. In Mediterranean regions, it frequently attacks vegetable plants: tomato, artichoke, Leguminoseae, and Cucurbitaceae, as well as tobacco, pink (*Dianthus*) and conifers. The caterpillar tends to be aggressive; it is carnivorous and subject to cannibalism.

The feeding larvae can be seen on the surface of plants, but are often hidden within plant organs (flowers, fruits, *etc*.). Bore holes and heaps of frass (excreta) may be visible, but otherwise it is necessary to cut open the plant organs to detect the pest. In temperate regions, it overwinters as a pupa buried several cm in the soil. Adults appear from April to May and can be observed until October due to their long migration period. Females lay several hundred eggs on all plant parts, including the flowers and fruits. Eggs may hatch in less than 3 days at an optimum temperature of 27 to 28˚C.

Plants are visually inspected for eggs and/or larvae to monitor and assess population sizes for *H. armigera*. In vegetative Australian cotton, a minimum of 60 whole plants per 100 ha of commercial fields are examined for the presence of *H. armigera* eggs or larvae; when plants begin to produce squares, only the upper terminal (approximately 20 cm) of a plant is inspected (Brown, 1984; Dillon and Fitt, 1995). In experimental plots, visual inspections for *H. armigera* in pigeon pea were restricted to the upper third of whole plants (4 sets of five plants in a 30x30 m plot) (Sigsgaard and Ersbøll, 1999). Leaves of tomato plants are more attractive than flowers or fruits of *H. armigera* oviposition sites, but the use of a single-leaf sample unit (with a sample size of 30 plants per field) has proven ineffective in detecting low densities of *H. armigera* (Cameron et al., 2001). On some tomato cultivars, leaves in the upper half of the plant are preferentially selected for oviposition (Saour and Causse, 1993). Larvae that are feeding on the surface of the plant are easily detected, but only entry holes or frass may be visible when larvae penetrate the plant; in this case, plant dissections are necessary in order to confirm the pest's presence (CABI, 2004).

Pheromone traps using (*Z*)-11-hexadecenal and (*Z*)-9-hexadecenal in a 97:3 ratio have been used to monitor populations of *H. armigera* (Pawar et al., 1988; Loganathan and Uthamasamy, 1998; Loganathan et al., 1999; Visalakshmi et al., 2000; Zhou et al., 2000). Of three pheromone doses tested in the field (0.75, 1.0, and 1.25 mg/septum), 1 mg attracted the most males (Loganathan and Uthamasamy, 1998); the trap type was not specified. Rubber septa impregnated with these sex pheromone components (1 mg/septum) were equally effective in capturing males for 11 days in the laboratory (Loganathan et al., 1999). Field captures of *H. armigera* were significantly lower with 15-day-old lures than with fresh lures; the authors recommend replacing lures every 13 days (Loganathan et al., 1999). Similar observations were reported by Pawar et al. (1988).

Trap design has a significant impact on the number of male *H. armigera* moths captured with pheromone lures. Funnel traps and Texas traps are substantially more effective than sticky traps (Kant et al., 1999). Hartstack (i.e., hollow cone) traps have also been used to effectively monitor densities of adults (Walker and Cameron, 1990). Cone traps are significantly more effective than water-pan traps (Sheng et al., 2002). Traps should be placed approximately 6 ft (1.8 m) above the ground (Kant et al., 1999; Zhou et al., 2000), and should be separated by a distance of at least 160 ft (50 m) (Kant et al., 1999). For routine monitoring of pests, pheromone traps are deployed at a density of 5 traps/ha (Sidde Gowda et al., 2002).

Adults of both sexes can be captured in black light traps.

Key Diagnostics

Several Noctuid pests can be confused easily with *Helicoverpa armigera*, including *H. assulta* [not known in the US]*, H. punctigera* [not known in the US]*, H. zea* [present in the US]*,* and *Heliothis virescens* [present in the US] (Kirkpatrick, 1961; CABI, 2000, 2003). Adults may be identified by distinct differences in genitalia (Kirkpatrick, 1961; Hardwick, 1965). A morphological study of *H. assulta, H. punctigera,* and *Heliothis virescens* (formerly *H. rubrescens*) compares similarities and differences between species; a key is provided for identifying adults (Kirkpatrick, 1961). Immunological tests are available to differentiate *H. punctigera* and *Heliothis virescens* in egg or larval stages (Ng et al., 1998).

The LepTon test, an Enzyme Linked Immunosorbent Assay (ELISA) based approach, has been developed to distinguish between *H. armigera* and *H. punctigera* in all stages (Trowell et al., 1993).

Phyllocnistis citrella

Scientific Name:
Phyllocnistis citrella Stainton

Synonyms:
Lithocolletis citricola, Phyllocnistis citricola

Common Name:
Citrus leafminer

Type of Pest:
Moth

Taxonomic position:
Class: Insecta, **Order:** Lepidoptera, **Family:** Gracillariidae

Reason for inclusion in manual
Added by National Science Program Leader, wounds often used for entrance of citrus canker bacterium

Pest description:
Eggs: Citrus leaf miner (CLM) eggs look like tiny droplets of water measuring only 0.3 x 0.2 mm. They are transparent when newly-laid, but become yellowish and opaque in 2 days. The eggs are laid singly on either surface of the leaf, but may also be laid on succulent stems. Newly-emerged leaflets, particularly along the midvein, are the preferred oviposition sites.

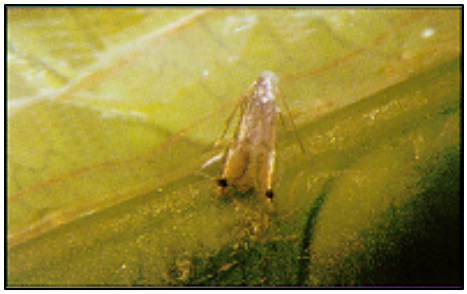


Figure 1. *P. citrella* adult. Photo courtesy of Texas A&M University – Kingsville Citrus Center and Texas Agricultural Experiment Station.

Larvae: The larvae are 1 to 2 mm long, light green and difficult to detect.

Pupae: The pupae are yellowish-brown, turning darker with age

Adults: Adults of CLM are minute moths (2 mm long with 4 mm wingspread) with white and silvery iridescent scales on the forewings, several black and tan

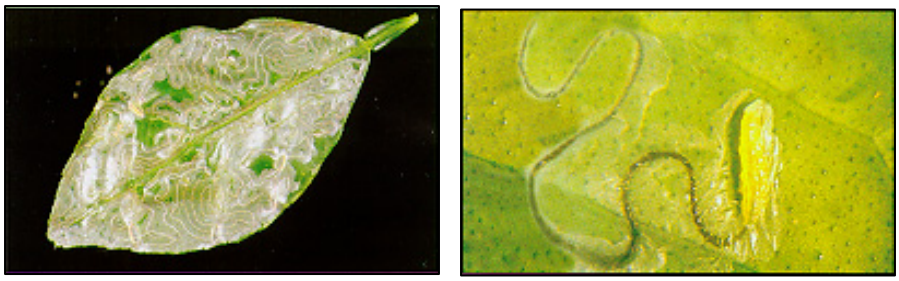


Figure 2. Silvery appearance of leaf mines (left) and serpentine, meandering leaf mines (right) of *P. citrella.* Photo courtesy of Texas A&M University – Kingsville Citrus Center and Texas Agricultural Experiment Station.

markings, plus a black spot on each wingtip (Fig. 1). The hind wings and body are white, with long fringe scales extending from the hindwing margins. The head is very smooth-scaled and white. The haustellum has no basal scales.

Biology and Ecology:
Total generation time for CLM ranges from 13 to 52 days, depending on temperature. The adults copulate 14 to 24 hours after emergence and lay their eggs soon afterwards. A single female may lay about 50 eggs during her lifetime. Larvae emerge from the eggs after 2 to 10 days. The larvae undergo four instars; the total development time takes 5 to 20 days. The first instar larvae begin feeding immediately, forming nearly invisible mines. During the course of larval development, the mines become more visible and larval excrement forms a central trail within the mine. When larval feeding is completed, the third instar usually forms a mine that is directed towards the leaf margin, where it molts into the fourth instar or prepupa. The prepupa is pale and almost cylindrical, and while it no longer feeds, it remains quite active. It forms a silken cocoon within the mine. As the silk dries, the leaf curls over the pupal cell. The pupal stage takes 6 to 22 days. Just before adult emergence, the pupa uses a spine on its head to make an opening at the anterior of the chamber, and forces its body through. Adults are most active from dusk to early morning; they spend the daytime resting on the undersides of leaves. Adults feed on nectar and live from 2 to 12 days.

Pest Importance
CLM is a pest of citrus species and other Rutaceae in many parts of the world. Heavy infestations can hinder the growth of newly planted trees or reduce fruit production of mature trees. Foliar and fruit damage can be extensive when populations of CLM are high. Although mining usually occurs on leaves, it also can occur on fruit, particularly grapefruit, reducing the market value of produce. CLM may also help spread citrus canker (Hill, 1918; Ando *et al.,* 1985) because of leaf damage from the mine.

Symptoms
CLM is easily detected by its meandering serpentine larval mine silvery in appearance and located on the leaf's ventral side (Fig. 2). Because the larvae generally do not cross existing mines, the later stages of feeding result in characteristically serpentine patterns. CLM larvae form mines predominantly in leaves, but also in succulent stems and sometimes fruit. The larvae bore through the leaf epidermis, ingesting the sap and producing a chlorotic leaf patch. CLM may prevent young leaves from expanding, causing them to remain twisted and curled. After the CLM have finished feeding, other insects, such as aphids and mealybugs, often continue feeding on the damaged area.

Known hosts
CLM attacks all citrus cultivars, many related to species in the Rutaceae family, and also some ornamentals. Grapefruit, tangerine and pummelo are among the most susceptible hosts. CLM has also been reported on cinnamon, jasmine, mistletoe, some legumes, willow and anaqua (*Ehretia anacua*).

Known distribution
The CLM is believed to have originated in India and southern China, where it is a major constraint to citrus production. In the last century, it has spread to the Philippines, Japan,

the Middle East, Australia and parts of western Africa. Its movement into northern Africa, Central America and the United States is recent. CLM was first reported in Florida in 1993. By late 1994, the invasion had spread to Alabama, Louisiana and Texas. CLM was found in Texas orchards (eastern Hidalgo County) in late August of 1994; by late November, it had spread throughout the southern Texas citrus growing area.

Potential distribution within the US

Citrus leafminer was found in late May 1993 in several citrus nurseries in Homestead, Florida, and Dade, Broward and Collier counties. It now occurs everywhere in Florida where citrus is grown, and has spread to other Gulf Coast areas. CLM is a New World, continental United States, and Florida resident. It is a potentially serious pest of citrus, associated Rutaceae, and some related ornamental plants (Beattie, 1989; Clausen, 1933; Kalshoven, 1981).

Survey

The best way to look for CLM is by visually examining citrus leaves. Young citrus leaves, especially the underside, will show evidence of a serpentine mine with a silvery appearance and curled leaves.

Larvae are minute, translucent greenish-yellow, and located inside the leaf mine. The pupae are in a pupal cell at the leaf margin. Adults are too minute to be easily noticed, and are active diurnally and in the evenings.

Spodoptera littoralis

Scientific Name
Spodoptera littoralis Boisduval

Common Name(s)
Cotton leafworm, Egyptian cotton leafworm, Mediterranean climbing cutworm, tobacco caterpillar, tomato caterpillar, Egyptian cotton worm, Mediterranean brocade moth, Mediterranean climbing cutworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera,
Family: Noctuidae

Reason for inclusion in manual
National Pest list and Eastern Region pest list

Figure 1. Eggs and neonates. Eggs are laid in in batches covered with orange-brown hair scales. Photo courtesy of http://www.defra.gov.uk/planth/pestnote/spod.htm

Pest Description
Eggs: Eggs are spherical, somewhat flattened, 0.6 mm in diameter, and laid in clusters arranged in, more or less, regular rows in one to three layers with hair scales derived from the tip of the female's abdomen (Fig. 1). Usually whitish-yellow in color, the change to a black color prior to hatching due to the larvae's head showing through the transparent shell (Pinhey, 1975).

Figure 2. (A). Larva of *S. littoralis*, showing light and dark longitudinal bands on sides of the body and dark spots on segments of the dorsal side (B) *S. littoralis* larva feeding on cotton leaves. Photo courtesy of CABI, 2004.

Larvae: Larvae grow to 40 to 45 mm and are hairless and cylindrical, tapering towards the posterior and variable in color (blackish-grey to dark green, becoming reddish-brown or whitish-yellow) (Fig. 2). The sides of the body have dark and light longitudinal bands. Their dorsal side has two dark semilunar spots on each lateral segment, except for the prothorax. Spots on the first and eighth abdominal segments are larger than others, interrupting the lateral lines on the first segment. The larva of *S. littoralis* is figured by Bishari (1934) and Brown and Dewhurst (1975).

Pupae: When newly formed, pupae are green with a reddish color on the abdomen, turning dark reddish-brown after a few hours (Fig. 3). The general shape is cylindrical, 14-20 x 5 mm, tapering towards the posterior segments of the abdomen. The last segment ends in two strong straight hooks (Pinhey, 1975).

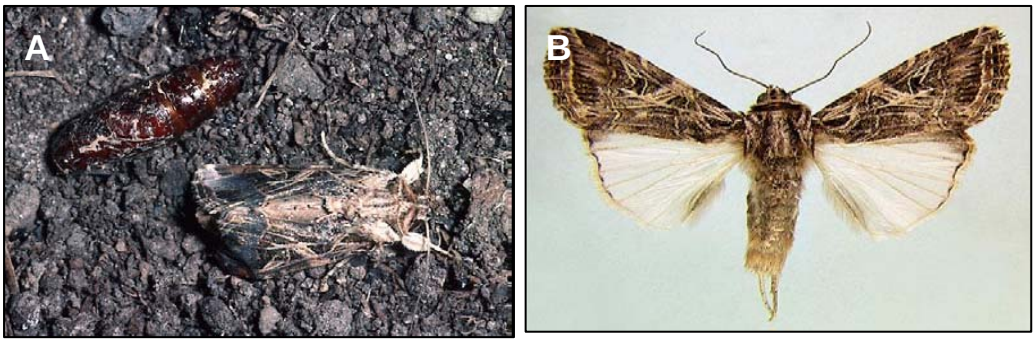


Figure 3. Pupa and adult of S. littoralis on soil (A). Adult moth of *S. littoralis* (museum set specimen) (B). Photos courtesy of CABI, 2004 and

Adults: Moths have a grey-brown body (Fig. 3), 15 to 20 mm long, and a wingspan of 30 to 38 mm with forewings grey to reddish brown with paler lines along the veins (in males, bluish areas occur on the wing base and tip). The ocellus is marked by two or three oblique whitish stripes. Hindwings are greyish white, irridescent with grey margins and usually lack darker veins (EPPO, 1997).

Pest Importance

Spodoptera littoralis is one of the most destructive agricultural lepidopterous pests within its subtropical and tropical range. It can attack numerous economically important crops throughout the year (EPPO, 1997). On cotton, the pest may cause considerable damage by feeding on leaves, fruiting points, flower buds and occasionally on bolls. When groundnuts are infested, larvae first select young folded leaves for feeding, but in severe attacks, leaves of any age are stripped off. Sometimes, even the ripening kernels in the pods in the soil may be attacked. Pods of cowpeas and the seeds they contain are often badly damaged. In tomatoes, larvae bore into the fruit, rendering it unsuitable for consumption. Numerous other crops are attacked, typically on their leaves.

In Europe, damage caused by *S. littoralis* was minimal until about 1937. In 1949, there was a catastrophic population explosion in southern Spain, which affected lucerne, potatoes and other vegetable crops. At present, this noctuid pest is of great economic importance in

Cyprus, Israel, Malta, Morocco and Spain (except the north). In Italy, it is especially important on protected crops of ornamentals and vegetables (Inserra and Calabretta, 1985; Nucifora, 1985). In Greece, *S. littoralis* causes slight damage in Crete on lucerne and clover. In north Africa, tomato, *Capsicum*, cotton, maize and other vegetables are affected. In Egypt, it is a serious cotton pest.

Symptoms

On most crops, damage arises from extensive feeding by larvae, leading to complete stripping of the plants. On cotton, the larvae feed on the leaves creating large holes of irregular shape with only the larger veins remaining. The larvae may also bore into the bud or young boll and consume the whole contents, causing them to be shed or dry up (Bishari, 1934). Bolls have large holes in them from which yellowish- to dark-green larval excrement protrudes. On tobacco, leaves develop irregular, brownish-red patches and the stem base may be gnawed off. Maize stems are often mined by *S. littoralis.* Young grains in the ear may be damaged.

Known Host

The host range of *S. littoralis* covers over 40 families, containing at least 87 species of economic importance (Salama et al., 1970). Economically important hosts include okra, onion, beet, cabbage, cauliflower, tea, bell pepper, watermelon, *Citrus spp*., coffee, carrot, cotton, soybean, fig, sunflower, sweet potato, potato, pea, bean, rice, tomato, cereal crops, tobacco, radish, roses, sugar cane, guava, spinach, cocoa, maize, cowpea, and grape vine.

Known Distribution

The northernly distribution limit of *S. littoralis* in Europe corresponds to the climatic zone in which winter frosts are infrequent. It occurs throughout Africa and extends eastward into Turkey and north into eastern Spain, southern France and northern Italy. This boundary is the extent of migrant activity as the pest overwinters in southern Spain, and not in northern Italy or France. In southern Greece, pupae have been observed in the soil after November and the species overwinters in this stage in Crete. Low winter temperatures are an important limiting factor affecting the northerly distribution, especially in a species with no known diapause (Miller, 1976; Sidibe and Lauge, 1977).

Potential distribution within the USA

The potential U.S. range of most spodoptera may be limited to the west coast through the lower southwestern and southeastern U.S., reaching as far north as Maryland. Migratory species may be capable of periodic spread into northern states and even Canada by late summer or early fall.

Survey (From Venette et al., 2003; CABI, 2004)

A number of sampling considerations for *S. littoralis* have been proposed. 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, skeletonizing these leaves as they feed on the surface. Sweep net sampling may be effective at dawn or dusk. Specimen identification should be confirmed by a trained taxonomist (USDA, 1982); however, not all sampling methods are equally effective for all insect life-stages. Eggs are only likely to be found by the 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. 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). Pheromone traps can be used to monitor the incidence of *S. littoralis* (Rizk et al., 1990). The synthetic sex pheromone (*Z*,*E*)-(9,11)-tetradecadienyl acetate is highly effective at trapping male moths of *S. littoralis* (Salem and Salama, 1985). Kehat and Dunkelblum (1993) found that the minor sex pheromone component, (9Z,12Z)-9,12-tetradecadienyl acetate, in addition to the major component (9Z,11Z)-9,11-tetradecadienyl acetate, was required for to attract males.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 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) 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., 1990). 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, and the densities of female *S. littoralis* affect the number of males captured at different times of the year (Rizk et al., 1990). 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 nondiscriminately capture multiple *Spodoptera* spp. (Blair, 1974) and other insects as well. A modified light trap using six 20-W fluorescent lights also proved effective for monitoring flight activity of *S. littoralis* (El-Mezayyen et al., 1997).

See http://www.aphis.usda.gov/ppq/manuals/pdf_files/NPRG-Spodoptera.pdf for additional survey information.

Key Diagnostics

Spodoptera littoralis is often confused with *S. litura.* The variability and similarity of the two species makes correct identification difficult; the examination of adult genitalia is often the only certain method available for species identification. For more information on the morphological discrimination between the adult, pupal and larval stages of the two species, refer to Schmutterer (1969), Cayrol (1972), Mochida (1973) and Brown and Dewhurst (1975). Although markings on larvae are variable, a bright-yellow stripe along the length of the dorsal surface is characteristic of *S. litura*. On dissection of the genitalia, the ductus and ostium bursae are the same length in female *S. littoralis*, but are different lengths in females of *S. litura*. The shape of the juxta in males, in both species, is characteristic, as is the ornamentation of the aedeagus vesica.

Thaumatotibia leucotreta

Scientific Name
Thaumatotibia leucotreta Meyrick

Synonyms:
Cryptophlebia leucotreta

Common Name(s)
False codling moth, citrus codling moth, orange codling moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for inclusion in manual
National Pest list and Eastern Region pest list

Pest Description
False codling moth, *T. leucotreta,* is an internal fruit feeding tortricid that does not undergo diapause. It is found throughout the year in warm climates on suitable host crops. *Thaumatotibia leucotreta* is a generalist with respect to host plant selection; it has been known to feed on over 50 different plant species. The generalist feeding strategy enables survival in marginal conditions as is necessary due to lack of diapause.

Eggs: flat oval shaped discs with a granulated surface and measurements varying from 0.77 mm in length by 0.60 mm in width up to 1 mm in diameter, the eggs are white to cream colored when initially laid, then changing to reddish color before the black head capsule of the larvae becomes visible under the chorion prior to eclosion (Daiber, 1979).

Larvae: first instar (neonate) larvae approximately 1 to 1.2 mm in length with dark pinacula giving a spotted appearance, fifth instar larvae are orangey-pink, becoming more pale on sides and yellow in ventral region, 12 to 18 mm long, with a brown head capsule and first thoracic segment (Fig. 1). The last abdominal segment bears an anal comb with 2 to 7 spines. The mean head capsule width (mm) for the first through fifth instar larvae has been recorded as: 0.22, 0.37, 0.61, 0.94 and 1.37, respectively (Daiber, 1979).

Figure 1. Larvae of *T. leucotreta.* Photo courtesy of T. Grove and W. Styn.

Pupae: Prepupa and pupa form inside a lightly woven silk and soil cocoon formed by the fifth

instar larvae on the ground. The length is 8 to 10 mm. Sexual determination through morphological differences on pupal case is possible (Daiber, 1979)

Adult: The adult body length ranges from 6 to 8 mm. Female and male wingspans range from 17 to 20 and 15 to 18 (mm), respectively. Antennae are setiform with distinct segments. Forewing coloration of the moth is similar between the sexes with gray, black, brown and orange-brown markings (CABI, 2004; Couilloud, 1988).

Biology and Ecology:

The adult moths emerge from cocoons located on the soil surface, mate and lay approximately 100 to 400 eggs at 15 and 25 °C, respectively, with very few eggs (0.4 per female) laid at 10 °C. Female moths undergo a pre-oviposition period of egg maturation, which averages 27 degree days above 12 °C. The onset and degree of oviposition vary with temperature and host plant. Peak oviposition occurs within three days after emergence and more than 50% of the eggs are laid in the first third of the reproductive period, which varies in length in an inverse relationship to temperature. Oviposition occurs at the highest rate in the early evening close to sunset.

Figure 2. Adult false codling moth. Photo courtesy of CABI, 2004.

Eggs can be laid singly on fruit or bolls of the crop. Neonate larvae emerge, wander the area and make an entry wound. Eggs can also be laid in small groupings of 2 to 4 “overlapping like tiles” on or near fruit surfaces. Eggs are only laid between 5pm and 10pm. Egg development takes 2 to 22 days, depending on temperature. Larvae typically complete five instars of development within a fruit or boll then exit and drop to the soil to begin construction of a cocoon. Pupation occurs on the soil surface, in the soil, in crevices under bark, in dropped fruit, or in debris. The empty pupal skin usually remains attached to the cocoon. Under laboratory conditions, the pupal stage lasts between 2 to 33 days, depending on temperature (Daiber, 1989). False codling moth is predominantly a warm climate pest and development is limited by cold temperatures. Exposure to temperatures below 10 °C reduces survival and/or development of several life stages; eggs have been killed at temperatures below 1 °C.

Pupae emerge slightly from the cocoon before adult emergence takes place. Emergence typically occurs in the early morning. Moths are active at night and spend daytime hours resting on shaded portions of the host. Moth activity increases with the onset of host flowering. Moths can mate several times per day. Oviposition occurs on or near developing fruit after petal fall. *Thaumatotibia leucotreta* females prefer specific parts of the host plant for oviposition. Females tend to choose smooth, non-pubescent surfaces for egg-laying. On cotton, green bolls are preferred. On peach, eggs are deposited near fruit on smooth leaves. Moths tend to select areas on fruit with damage.

Pest Importance (from Venette et al., 2003)

Thaumatotibia leucotreta is a pest of economic importance to several crops, including corn, cotton, citrus, litchi, macadamia, peach and plum throughout sub-Saharan Africa, South Africa, and the islands of the Atlantic and Indian Oceans. Larval feeding and development can affect fruit development at any stage, causing premature ripening and fruit drop.

All stages of citrus and stone fruits are vulnerable to attack (Fig. 3). *Thaumatotibia leucotreta* larvae are capable of developing in hard green fruit before control measures can be started. Once a fruit is damaged, it becomes vulnerable to fungal organisms and scavengers. In peaches, there is up to 28% loss of late-peach crops (CABI, 2004). Larvae damage stone fruits as they burrow into the fruit at the stem end and begin to feed around the stone. Detecting infested peaches can be difficult if the fruit is still firm and abscission has not occurred; consequently, the danger of selling potentially infested fruit poses a serious threat to the peach industry. On oranges, *T. leucotreta* caused 2 to 5% damage on Valencia and Navel oranges in 1954, but yield losses have been as great as 10 to 20% (CABI, 2004). An infested orange shows brown, sunken spots with larval holes bored in the center of the spot.

Figure 3. A *T. leucotreta* larva in an orange fruit. Photo courtesy of N. Sishuba. Rhodes University, South Africa.

Thaumatotibia leucotreta has caused significant yield losses (≥30%) to macadamia crops in Israel and South Africa. Damage to macadamia nuts is caused from larvae feeding on the developing kernel after they pierce the husk and shell. In Ugandan cotton, *T. leucotreta* caused 20% loss of early sown varieties and 42 to 90% loss of late varieties. Larval penetration of cotton bolls facilitates entry of other microorganisms that can rot and destroy the boll.

Symptoms

Symptoms vary according to host. On oranges, there is sometimes a scar on the fruit surface. On most other crops, the habit of internal feeding leaves few symptoms. Emerging larvae bore into the albedo and usually feed just below the fruit surface. Cannibalism among young larvae ensures that usually only one caterpillar matures in each fruit. When full-grown, the larvae bore their way out of the fruit to seek a site for pupation. The rind around the point of infestation takes on a yellowish-brown color as the tissue decays and collapses. Larval feeding and development can affect fruit development at any stage, causing premature ripening and fruit drop. The degree of damage is highly variable from orchard to orchard and from season to season, and can be as high as 90% of a citrus crop.

Known Hosts

False codling moth feeds on more than 70 host plants (CABI, 2004). Economically important hosts include avocado, banana, bean, cacao, carambola, castorbean, citrus (*Citrus sinensis*, *Citrus spp.*), coffee, corn (*Zea mays*), cotton, cowpea, English walnut, grape, guava, macadamia nut, mango, okra, olive, peach , pepper/pimento, persimmon,

plum, pineapple, pomegranate, sorghum, and tea (Venette et al., 2003; CABI., 2004).

The navel cultivar appears to be the variety of citrus most heavily attacked by the false codling moth. Grapefruits and mandarins are less susceptible, and larval development is rarely, if ever, completed in lemons and limes, which could be the result of their greater acidity and excessive juiciness.

Known Distribution

Thaumatotibia leucotreta has a large distribution in Africa, but appears to be fairly limited in other parts of the world.

Potential Distribution within the US

No wild infestations of *T. leucotreta* have been reported in the US. The apparently moderate rate of arrival combined with the potentially limited availability of suitable climate lowers the likelihood of establishment. Because this pest has a broad host range and suitable host plants are both common and abundant, a relatively high probability of pest establishment exists if introduced into a suitable climate. Should this pest become established in the US, the economic consequences are likely to be severe (Venette et al., 2003).

Survey (From Venette et al., 2003)

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

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). Mohr (1973) speculates that pheromones 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. These components are most effective when used in a ratio between 70:30 and 30:70 (E:Z). 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. 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, but these have not performed as well as the Hoechst Biotrap or Pherocon 1C traps. 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). Lures should be replaced every 8 weeks. Traps should be placed approximately 5 ft (1.5m) high. For routine monitoring, 1 to 2 traps per acre (2 to 5 traps/ha) 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. Lures for *T. lecutreta* can be used in the same trap with lures for *P. scutigera*. Pheromone lures with (E)- and (Z)-8-dodecenyl acetate may also attract *Cydia cupressana* (native), *Hyperstrotia spp*. (PPQ, 1993), Cydia *atlantic*a (exotic), *Cydia phaulomorpha* (exotic) and *Cryptophlebia peltastica* (exotic).

In citrus, attempts to disrupt mating in *T. leucotreta* with the two-component pheromone blend successfully disoriented males but failed to reduce damage caused by larvae.

Key Diagnostics

Thaumatotibia leucotreta can be confused with many *Cydia* spp., including *C. pomonella* (codling moth), because of its similar appearance and damage; however, unlike codling moth, its host range does not include apples, pears or quince (USDA, 1984). In west Africa, *T. leucotreta* is often found in conjunction with *Mussidia nigrevenella*; however, they can be distinguished by close examination of morphological characters (CABI, 2004). In South Africa, there is also an overlapping host range for *T. leucotreta* and *Cydia peltastica*, particularly on litchi and macadamia (Venette et al., 2003).

Psyllids

Diaphorina citri

Scientific Name
Diaphorina citri Kuwayama

Synonyms:
Euphalerus citri

Common name(s)
Asian citrus psyllid, Asiatic citrus psyllid, oriental citrus psyllid

Type of Pest
Psyllid

Taxonomic Position
Class: Insecta, **Order:** Homoptera, **Family:** Psyllidae

Reason for inclusion in manual
Western Region pest list

Pest Description
The Asian citrus psyllid, *Diaphorina citri*, is similar to *Trioza erytreae*, the African citrus psyllid, which is the vector of citrus greening disease in Africa. The geographical range of the two species did not originally overlap, but they now occur together in Mauritius, Reunion, and Saudi Arabia (CABI, 2004).

Figure 1. Eggs of *D. citri*. Photo courtesy of D. Caldwell.

Eggs: The egg of *D. citri* is anchored on a slender stalk-like process arising from the plant tissue. It is elongate with a broad basal end and tapering towards its distal and curved end. The average size of the egg measures 0.31 mm long and 0.14 mm wide. Freshly deposited eggs are light yellow and turn bright orange with two distinct red eye spots at maturity (Fig. 1) (CABI, 2004). Eggs are placed on plant tissue with long axis vertical to surface (long axis is horizontal to surface in *T. erytreae*) (Mead, 2002). Females can lay more than 800 eggs during their lives.

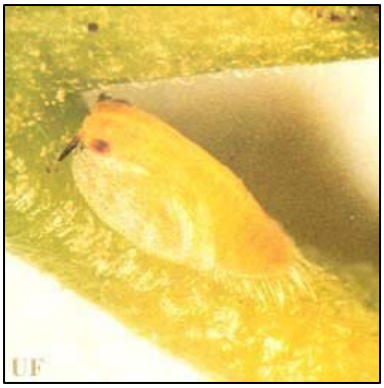


Figure 2. Nymph of *D. citri*. Photo courtesy of University of Florida.

Nymph: The average size of first-instar nymphs is 0.30 mm long and 0.17 mm wide. The nymphs have a light pink to yellowish orange body and a pair of red compound

eyes (Fig. 2). Second-instar nymphs are on average 0.45 mm long and 0.25 mm wide. The rudimentary wing pads are visible on the dorsum of the thorax. The average size of third-instar nymphs is 0.74 mm long and 0.43 mm wide. The wing pads are well developed and the segmentation of antenna is evident. The fourth-instar nymphs average 1.01 mm long and 0.70 mm wide. The wing pads are well developed; the mesothoracic wing pads extend towards the one-third of compound eyes and the metathoracic wing pads extend to the third abdominal segment. The fifth-instar nymphs average 1.60 mm long and 1.02 mm wide. The mesothoracic wing pads extend towards the front of the compound eyes; the metathoracic wing pads extend to the fourth abdominal segment. In some mature nymphs, the abdomen turns bluish-green instead of pale orange (CABI, 2004). There are no abdominal spots present (advanced nymphs of *T. erytreae* have two basal dark abdominal spots). Wing pads are massive (small pads in *T. erytrea*e) and large filaments are confined to apical plate of abdomen (*T. erytreae* with fringe of white filaments around whole body, including head) (Mead, 2002).

Adult: 2.5 mm long, body mottled brown, legs greyish-brown, head light brown (black in *T. erytreae*) (Fig. 3). Wings have a dark pigmented band around the edges that has a break just below the distal tips. There are scattered pigmented spots in the center of the wings. Apical half of wing is broadest. The average size of adult females is 3.3 mm long and 1 mm wide; the mean size of of adult males is 2.7 mm long and 0.8 mm wide. *T. erytreae* wings are unspotted, broadest in the middle, and pointed at the tips (CABI, 2004; Mead, 2002). *T. erytreae* nymphs live in individual pitgalls on the underside of leaves, whereas *D. citri* nymphs are free-living. The living insect is covered with whitish, waxy secretion, making it appear dusty.

Figure 3. Adult *D. citri*. Photo courtesy of D. Caldwell.

Biology and Ecology

Diaphorina citri has a short life-cycle and high fecundity. It is more prevalent in hot coastal areas. Pairing starts soon after emergence. The insects are most active during March through April in India, May through June in the Philippines, and June through July in Brazil. Gravid females of *D. citri* oviposit within 2-cm lengths of the terminal tissue in leaf folds, on petioles, axillary buds, upper and lower surfaces of young leaves and tender stems. The average incubation period of eggs at 25°C ranges from 4.1 to 4.3 days. All nymphs undergo at least four molts. The first- and second-instar nymphs typically aggregate and feed on the inside of folded leaves, on the terminal stem, and between the axillary bud and the stem of tender shoots. Young nymphs are quite docile and move only when disturbed or over-crowded. The nymphs continuously secrete copious amounts of honeydew from the anus and a thread-like waxy substance from the circumanal glands resulting in the growth of black sooty mold on the lower leaves. The average combined developmental periods for the five nymphal stages are 12.8, 12.6, 13.5, and 13.1 days on orange jasmine, grapefruit, rough lemon, and sour orange, respectively (Tsai et al., 2002).

Adults of *D. citri* often are found resting on the terminal portion of the plant, especially on the lower side of the leaves with their heads either pointing upward or downward to the leaf surface at an angle of 30°. When disturbed, they readily take flight for a short distance. The

females only oviposit on the tender shoots. In the absence of suitable host tissue, oviposition temporarily ceases. The longest female longevity of *D. citri* reared on grapefruit, orange jasmine, sour orange, and rough lemon at 25°C was 54, 54, 60, and 66 days, respectively (CABI, 2004). In an insectary, at 10, 15, 20, 25, 28, and 33°C, the psyllid populations reared at 10°C and 33°C failed to develop.

The optimum range of temperatures for population growth of *D. citri* is 25 to 28°C. During dry periods, adults are numerous, but nymphs are usually absent. The complete life-cycle takes 14 to 48 days, with up to 10 overlapping generations per year. The adults overwinter and can live for up to 6 months. Population fluctuations are closely correlated with the flushing rhythm of citrus trees, as eggs are laid exclusively on young flush points. In southern Florida, *D. citri* populations occur throughout the seasons on orange jasmine, but there are population peaks in October through November, December, May, and August, which are positively related to the weekly minimum temperature and rainfall.

Pest Importance

The Asian citrus psyllid is widely distributed in southern Asia. It is a pest of citrus in several countries, particularly India, where there has been a serious decline of citrus in recent years. This psyllid did not occur in North America or Hawaii, but was reported in Brazil by Costa Lima (1942; Rio de Janeiro) and Catling (1970). In June 1998, the insect was detected in Florida along Highway 1 on the east coast from Broward to Martin counties; it was apparently limited to dooryard host plantings at the time of its discovery. By September 2000, this pest had spread to 31 counties in Florida (Halbert, 2001), mostly by the sale of *Murraya paniculata* in discount outlets. *Diaphorina citr*i, and one of its parasites, is also present in the Rio Grande Valley of Texas. Both species appear to have been accidentally introduced in the spring of 2001 on potted *Murraya* originating in Florida.

Diaphorina citri is the vector of a serious citrus greening disease caused by the bacterium *Candidatus* Liberibacter asiaticus. *Diaphorina citri* transmits the Asian form of citrus greening disease under natural conditions in Asia (including Saudi Arabia). Through experimental data, *D. citri* is known to transmit the African form of the disease, *Candidatus* Liberibacter africanus. In Mauritius and Réunion, where both forms occur, *D. citri* probably transmits both forms. *Diaphorina citri* is the likely vector of *Candidatus* Liberibacter americanus in Brazil.

Diaphorina citri travels short distances. Citrus material (grafted trees, rootstock seedlings) from infected areas can carry eggs and/or nymphs over longer distances. *Candidatus* Liberibacter asiaticus probably was introduced into Saudi Arabia via infested plant material. The rutaceous plant *Murraya paniculata*, frequently used as an ornamental bush or hedge, is one of the best hosts of *D. citri*. It can carry eggs or nymphs of the vector; its introduction to disease- and vector-free regions could therefore be dangerous. *Murraya paniculata* is susceptible to *Ca*. L. americanus, but it is not susceptible to strains in Taiwan. *D. citri* has been intercepted regularly on

Figure 4. Distortion of leaves. Photo courtesy of EPPO.

fruit sent from the Bahamas to Florida for processing.

Symptoms/Signs

Injury caused by psyllids results in leaf distortion caused by nymphal feeding, heavy development of sooty mold on honeydew-covered leaves, and transmission of the organisms that cause greening disease (Mead, 2002). *Diaphorina citri* damages citrus by injecting a salivary toxin that produces malformation of shoots and leaves (Fig. 4) (Tsai et al, 2002).

Serious damage to growing points can occur occasionally, which can lead to dwarfing as well as taste abnormalities in fruit due to citrus greening disease. Heavy *D. citri* populations can cause blossom and fruitlet drop. The honeydew excreted by *D. citri* promotes the growth of sooty mold, which not only affects the photosynthetic activity of the tree, but also attracts ants that fend off natural enemies of *D. citri,* resulting in additional pest damage. *Diaphorina citri* stunts and twists young shoots, so that the growing tips present a rosetted appearance. Leaves are often badly curled (CABI, 2004; Mead, 2002).

The white waxy excretions of the nymphs are an indicator of *D. citri* (Fig. 5). In heavy infestations, eggs are so numerous that the newest growth (1-5mm) on terminal foliage has an orange appearance.

Figure 5. White waxy excretions of *D. citri* nymphs. Photo courtesy of D. Caldwell.

Known Hosts

Diaphorina citri is confined to the Rutaceae, occurring on wild hosts, as well as on Citrus, especially lemons (*C. limon*), rough lemon (*C. jambhuri*), sour orange (*C. aurantium*), grapefruit (*C. paradisi*), and limes (*C. aurantiifolia*). *Murraya paniculata*, a rutaceous plant often used for hedges, is a preferred host. *Murraya koenigii* is a host in India and Sri Lanka (CABI, 2004)

Known Distribution

Diaphorina citri ranges in tropical and subtropical Asia; it has been reported in the following geographical areas: Bangladesh, Bhutanm Cambodia, China, India, Myanmar, Taiwan, Philippine Islands, Malaysia, Maldives, Indonesia, Singapore, Sri Lanka, Pakistan, Thailand, Nepal, Cecum, Hong Kong, Vietnam, Yemen, Japan (Ryukyu Islands), Afghanistan, Saudi Arabia, Iran, Paraguay, Uruguay, Venezuela, Reunion, Mauritius, Argentina, Brazil, Bahamas, Cuba, Guadeloupe, and the United States (Florida, Texas, Hawaii) .

The distribution of *D. citri* is wider than that of the citrus greening bacterium *Candidatus* Liberibacter asiaticus, the major pathogen that it transmits. *Diaphorina citri* occurs in Guadeloupe, Honduras, Hong Kong, Iran, Macau, Singapore, Sri Lanka, Florida (USA) and South America, where the bacterium has not been recorded.

Potential distribution within the US

Currently present in Florida, Texas, and Hawaii.

Survey (from CABI, 2004)

Murraya paniculata, an ornamental that is widely grown in Florida, is a preferred host of *D. citri*. Populations on this plant can be extremely high. Inspection of this plant may be the best way to survey for the Asian citrus psyllids (Halbert, 1999).

Sooty mold on foliage indicates the presence of Sternorrhyncha (which can also be due to scales and aphids).

Nymphs: The nymphs are always found on new growth, and move in a slow, steady manner when disturbed. The white waxy excretions of the nymphs are an indicator of *D. citri*. Also, a single *D. citri* nymph feeding for less than 24 hours on a citrus leaf induces a distinctive and permanent deformation that becomes increasingly evident as the leaf expands. This characteristic damage is a useful tool for estimating psyllid activity because it remains evident long after nymphs have been eliminated by predation or other sources of mortality. There are no galls or pits formed on the leaves, as caused by many other kinds of psyllids; the nymphs are completely exposed.

Adults: The adults will leap when disturbed and may fly a short distance. They are usually found in large numbers on the under sides of the leaves with heads almost touching the surface and bodies raised almost to a 30 degree angle. The period of greatest activity of the psyllid corresponds with the periods of new growth of citrus.

In a study to examine the seasonal abundance of *D. citri* on orange jasmine (*M. paniculat*a) and grapefruit (*C. paradisi*), Tsai et al. (2002) took random samplings for psyllid adults at weekly intervals. One shoot (6 to 10 cm long) was selected from every square meter area by randomly throwing a pointed object made of a pencil with a ribbon tied to one end. The shoot was selected where the pencil had landed. A total of 100 shoots were selected from each field on each sampling date. Dharajothi et al. (1989) collected ten new shoots (4 to 5 cm long), which were sampled at random from each cardinal direction (East, West, North, and South) for a total of 40 shoots per tree. The shoots were cut and collected in glass tubes and brought to the laboratory for population estimation. The egg and nymph populations were counted by observing shoots under a binocular dissection microscope. Adult populations were counted in the field.

Field recognition of greening in Asia from symptoms alone is often difficult to recognize (see citrus greening). Greening symptoms of citrus include stunted growth, sparsely foliated branches, unseasonal bloom, leaf and fruit drop, and twig dieback. Young leaves are chlorotic, with a blotchy mottle that does not follow veins. Mature leaves have irregular, yellowish-green patches and midribs that are usually yellow. In severe cases, leaves become chlorotic and have scattered spots of green. Fruits on greened trees are small, generally lopsided, underdeveloped, unevenly colored, hard, and poor in juice and taste. The columella was found to be almost always curved in sweet orange fruits and the most reliable diagnostic symptom of greening. Most seeds in diseased fruits are small and dark colored. Most fruit falls off the tree prematurely.

Key Diagnostics

Identifications having regulatory significance should be made by taxonomists with adequate

reference materials. Adult psyllids are most likely confused with leafhoppers; nymphs resemble scales. Differences with the African citrus psyllid are given within the text of this document where appropriate.

Trioza erytreae

Scientific Name:
Trioza erytreae (Del Guercio)

Synonyms
Aleurodes erytreae, *Spanioza eritreae*, *S. erythreae*, *S. erytreae*, *Spanioza merwei, T. citri*, *T. erythreae*, *T. merwei*

Common Names
African citrus psyllid, African citrus psylla, citrus psylla (African), citrus psyllid (African), two-spotted citrus psyllid

Type of Pest
Psyllid

Taxonomic Position
Class: Insecta, **Order:** Hemiptera, **Family:** Triozidae

Reason for inclusion in manual
National CAPS List, Western Region pest list

Pest Description
Trioza erytreae is similar to *Diaphorina citri*, the Asian citrus psyllid, which is the vector of citrus greening in Asia. The geographical range of the two species did not originally overlap, but they now occur together in Mauritius, Reunion and Saudi Arabia (CAB, 2004).

Eggs*:* Orange, cylindrical, with a sharp point anteriorly (Fig. 1); laid on leaf margins of young, actively growing foliage. Eggs are placed on plant tissue with long axis horizontall to surface (long axis is horizontal to surface in *D. citri*).

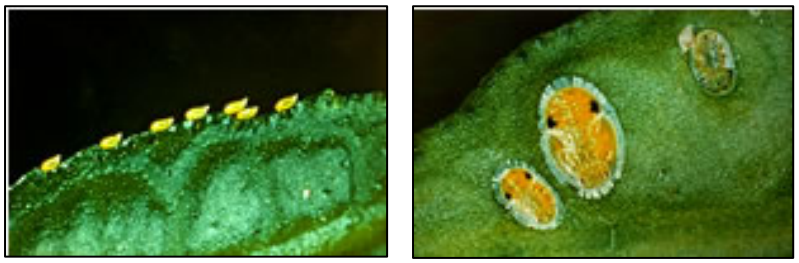


Figure 1. Eggs (left) and nymphs (right) of *T. erytreae.* Photo courtesy of EPPO.

Nymph: Dorso-ventrally compressed and varying in color from yellow, olive-green to dark grey; has a marginal fringe of white, waxy filaments; largely sedentary; forms distinct colonies and settles on the underside of young leaves, where, after a few days of feeding, it produces distinctive cup-shaped, open pit galls. Late instar nymphs of *T. erytreae* have two basal dark abdominal spots (Fig. 1).

Adult: Winged, pale and delicate initially, later becoming light brown. Males are smaller than females and have a turned up abdomen, the latter ending in a sharp point in females.

When feeding, adults take up a distinctive stance, with the abdomen raised at an angle of about 35° to the feeding surface.

Biology and Ecology

Trioza erytreae has a temperature sensitivity (does not develop at temperatures above 77 ºF) similar to that of Candidatus *Liberibacter africanus* (the agent of citrus greening in Africa). It is sensitive to hot, dry weather (the eggs and first-instar nymphs being particularly vulnerable). It favors cooler, moist areas above 500 to 600m in altitude, where citrus growth flushes tend to be prolonged.

Sex ratios fluctuate in the field, but females always predominate. There is a pre-oviposition period of 3 to 7 days, but this is considerably extended in the absence of young foliage. Mating occurs 2 to 4 times a day and eggs may be laid immediately. Eggs are supplied with a sharp point that is driven through the leaf epidermis and is thought to be responsible for maintaining a favorable internal water relationship. Females remain fertile for 11 to 16 days in the absence of males, and maximum egg production occurs towards the middle of their life span, which normally lasts 17 to 50 days; up to 2000 eggs may be laid per female. There is an incubation period of 6 to 15 days; nymphal development (five instars) takes 17 to 43 days, both periods being inversely related to mean temperature and directly related to nutritional value of the leaves. The temperature threshold for nymphal development is around 10 to 12°C with no diapause (CABI, 2004).

Pest Importance

Trioza erytreae is the vector of the very serious citrus huanglongbing (greening) disease caused by Candidatus Liberibacter species (EPPO/CABI, 1997). Heavy infestations of *T. erytreae* alone, however, can cause severe leaf distortion and the development of conspicuous pits on the leaf surface (Fig. 2).

Like the other vector of citrus greening (*Diaphorina citri*), *T. erytreae* is listed as an A1 quarantine pest by EPPO (OEPP/EPPO, 1988), and is also a quarantine pest for CPPC and OIRSA. It is primarily a pest in tropical climates and, as such, could become established in citrus-growing countries in the Americas and Asia. It could probably establish and spread without difficulty in citrus-growing countries within the Mediterranean region. Besides its role in the spread of citrus huanglongbing disease, the psyllid itself has significant damage potential.

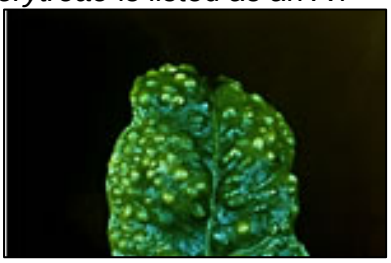


Figure 2. Leaf galls of *T. erytreae*. Photo courtesy of EPPO.

Trioza erytreae transmits the causal agent of the African form of citrus huanglongbing (greening) disease, *Candidatus* Liberibacter africanus, under natural conditions in Africa and the Middle East (McClean and Oberholzer, 1965). It has been experimentally shown that *T. erytreae* is able to transmit the Asian form, Candidatus Liberibacter asiaticus (Massonie et al., 1976).

Symptoms

Trioza erytreae severely distorts leaves, which have become stunted and galled (Fig. 2), and appear dusted with fecal pellets. Young leaves may be yellow in color.

Known Hosts
Trioza erytreae is confined to the family Rutaceae, occurring on wild hosts (e.g., *Clausena anisata*) and*Citrus spp.*

Known Distribution
*Trioza erytrea*e has been reported in Saudi Arabia, Yemen, Portugal, Madeira, Cameroon, Comoros, Congo Democratic Republic, Eritrea, Ethiopia, Kenya, Madagascar, Malawi, Mauritius, Rwanda, Saint Helena, Somalia, South Africa, Sudan, Swaziland, Tanzania, Uganda, Zambia, and Zimbabwe (EPPO, 2004).

Potential Distribution in the US
The African form of citrus greening develops only under cool temperatures of 68 to 77 ºF, which is mostly due to the temperatures required for vector development. Therefore, the potential distribution within the US would include areas with Citrus production and relatively cool temperatures.

Survey
Trioza erytreae can be easily detected on citrus leaves, due to the nymphs being in pitlike galls that accommodate the developing insects. Since citrus leaves remain on the tree for two or more seasons, a count of the leaves pitted by psylla nymphs indicates previous *T. erytreae* activity. *Trioza erytreae* can be detected in the field with the naked eye; however, identification of immatures requires the use of a hand lens or similar magnification. External foliage of the citrus tree should be examined for the presence of symptoms (described above) that could potentially signal the presence of the pest. Adults and immatures should be collected and preserved in 80% ethanol for taxonomic verification.

Because indigenous plants of the family Rutaceae host *T. erytrea*e, it is essential to detect wild reservoirs of the insect pest in citrus areas. Monitoring traps can be positively phototaxic of the responding optimally to wavelengths 500 to 550 nanometers (yellow-green) (Aubert, 1987). The attractive component of the trap can be a 3M Saturn Yellow Adhesive tape. Sticky traps can attract many other species of *Trioza* which are very similar in appearance to *T. erytreae.* The traps should only be used if *T. erytreae* is known to occur in high numbers in the absence of other *Trioza* species. Extremely high numbers of *T. erytreae* occur in some rainforest areas of Cameroon on isolated citrus orchards and nurseries, and have resulted in catches of 2 to 3 winged adults per square cm of tape per day; however, psylla populations are generally much lower in African orchards (Aubert, 1987).

Key Diagnostics
Identifications having regulatory significance should be made by taxonomists with adequate reference materials. Adult psyllids are most likely confused with leafhoppers; nymphs resemble scales. Differences with the Asian citrus psyllid are given within the text of the *Diaphorina citri* document.

Scale Insects

Ceroplastes destructor

Scientific Name
Ceroplastes destructor Newstead

Common Name(s)
White wax scale, citrus waxy scale, soft wax scale, white scale, white waxy scale

Type of Pest
Scale insect

Taxonomic Position
Class: Insecta, **Order:** Hemiptera, **Family:** Coccidae

Reason for inclusion in manual
National pest list and Eastern Region pest list

Pest Description
There are three nymphal instars. Early instars are morphologically difficult to distinguish from other *Ceroplastes* species. Wakgari and Giliomee (1998) provide a key to different stages of *C. destructor* and detailed descriptions and illustrations of all nymph and adult stages. The following morphology is taken mainly from Wakgari and Giliomee (1998) for the nymph stages and from Qin and Gullan (1994) and Wakgari and Giliomee (1998) for the adult. These descriptions include details visible only in specimens mounted on microscope slides, examined under high magnification.

First-instar nymph: The first-instar nymph is oval, dorsolaterally flat, 0.32 to 0.50 mm long; eye-spots heavily pigmented, present dorsolaterally on each side of the head region; marginal setae flagellate, 9 to 12 between anterior spiracular furrows, 2 to 3 between anterior and posterior spiracular furrows, 7 between posterior spiracular furrow and anal cleft; three spiracular setae present in each spiracular furrow; dorsum without setae or pores; anal plates each with a very long, slender apical seta and three other dorsal setae, one fringe seta and one ventral seta; venter without submarginal setae, one pair of interantennal setae and one pair of prevulvar setae present, a few cruciform pores present in submargin, three quinquilocular pores between spiracle and its corresponding furrow; antennae 6-segmented; legs well developed, without tibiotarsal sclerosis, tarsal digitules equal in size and knobbed, claw denticle absent, claw digitules unequal, one slender and one stout, both apically knobbed.

Second-instar nymph: The second instar is oval, 0.65 to 0.70 mm long, eye-spots pigmented, present dorsolaterally on each side of the head region; marginal setae flagellate, 8 to 10 between anterior spiracular furrows, 2 between anterior and posterior spiracular furrows, 6 to 7 between posterior spiracular furrow and anal cleft; three spiracular setae present in each spiracular furrow; dorsum without setae, with some bilocular pores

along submarginal areas; anal plates each with four dorsal setae, two fringe setae and one ventral seta; venter with bristle-shaped submarginal setae, two pair of interantennal setae (one longer one shorter) and one pair of prevulvar setae present, cruciform pores present in submargin, four to seven quinquilocular pores between spiracle and its corresponding furrow; antennae 6-segmented; legs well developed, similar in structure to that of first-instar nymph.

Third-instar nymph: The third instar is oval, 0.85 to 1.20 mm long, eye-spots black, present dorsolaterally on each side of the head region; marginal setae flagellate, occasionally clavate to capitate, 9 to 14 between anterior spiracular furrows, 3 to 5 between anterior and posterior spiracular furrows, 7 to 11 between posterior spiracular furrow and anal cleft; 6 to 10 spiracular setae present in each spiracular furrow; dorsum with one anterior and six lateral dorsal clear areas, setae and pores sparsely distributed over dorsum except lacking in dorsal clear areas, pores bilocular, oval trilocular or triangular trilocular and a few monocular in median area; anal plates each with four dorsal setae, three fringe setae and one ventral seta; venter with bristle-shaped submarginal setae and sparsely distributed ventral setae, two pairs of interantennal setae (one longer, one shorter) and one pair of prevulvar setae present, cruciform pores present in submargin around entire body, 12 to 18 quinquilocular pores between spiracle and its corresponding furrow; antennae 6-segmented; legs well developed, similar in structure to that of first-instar nymph but relatively smaller compared with body size.

Adult female: After Qin and Gullan (1994) and Wakgari and Giliomee (1998). There is some variation in the number of pores and setae recorded by Qin and Gullan (1994) and Wakgari and Giliomee (1998). This result is due to Qin and Gullan's (1994) use of Australian material, with counts including both young and old specimens, while Wakgari and Giliomee (1998) made use of African material, with counts including only young specimens.

Adult females are oval, sometimes with some marginal indentation, 2.5 to 6.40 mm long, with a strongly sclerotized anal process; eye-spots black, relatively small, present dorsolaterally on each side of the head region; marginal setae bristle-like, 8 to 24 between anterior spiracular furrows, 4-8 between anterior and posterior spiracular furrows, 7 to 12 between posterior spiracular furrow and anal cleft; 37 to 77 spiracular setae present in each spiracular furrow; dorsum derm membranous in young specimens but becoming sclerotized in old specimens, with one anterior and six lateral dorsal clear areas, dorsal setae cylindrical, some with apex slightly expended, evenly distributed over dorsum except lacking in dorsal clear areas, dorsal pores oval trilocular or triangular trilocular and occasionally quadrilocular or bilocular, preopercular pores numbering 5 to 6 in single transverse row immediately anterior to anal plates; anal plates each with four dorsal setae, four fringe setae and one ventral seta; venter with submarginal setae and sparsely distributed ventral setae, two pair of interantennal setae (one longer one shorter) and one pair of prevulvar setae present, cruciform pores mostly present in submargin around entire body, sparse in other ventral areas, 65 to 110 quinquilocular (a few with 6 or 7 loculi) pores between spiracle and its corresponding furrow; multilocular pores distributed around vulva and in a band across preceding segment, mostly with 10 loculi, tubular ducts present on abdomen; antennae 6-segmented; legs well developed, similar in structure to that of first-instar nymph but relatively much smaller compared with body size.

The adult male is unknown.

Pest Importance

Ceroplastes destructor was once a major pest of citrus in New South Wales, Queensland and Western Australia. Documents with regard to insect pests of these states in the 1950s, 1960s and 1970s were devoted to examining the life history and appearance of the insect, in addition to methods for its control using oil and insecticidal sprays. It is now a minor pest in Australia (Smith et al., 1997) due to the control of natural enemies from Africa. *Ceroplastes destructor* has been a minor pest in native African countries because there is a complex of natural enemies that have been kept under control for over 3 decades. Recently, *Citrus reticulata* (Blanco) has become important to the Western Cape Province of South Africa (Wakgari and Giliomee, 1998).

Symptoms

Ceroplastes destructor attacks the leaves, branches and stems of host plants, which will affect its vigour and growth. A large number of young crawlers can be seen on leaves when the eggs hatch, but these do not persist. They usually settle on the leaf surface along midribs or leaf petioles. Once the crawlers settle down, they start secreting white wax (Fig 1). Gimpel et al. (1974) describes, in detail, the process and shapes of the wax produced by wax scales; *C. destructor* produces wax in a similar way. After 3 to 4 days of settlement, the dorsal wax pad appears as a thin, white marking. The wax rays gradually appear around the body margin. The insects move from their original settlement site to the twigs at the beginning of the third instar. At this stage, the wax builds up like a cone and, when more wax is secreted, the late third instar attains its characteristic oval shape. The adults are completely covered with white wax in irregular shapes. Qin and Gullan (1994) and Wakgari and Giliomee (1998) contain figures showing the wax appearance of the different stages. Sooty mold is usually associated with the scales.

Figure 1. *C. destructor* on coffee. Females are immobile and covered in a white, waxy layer. Photo courtesy of CABI, 2004.

Known Hosts

Ceroplastes destructor is polyphagous and attacks a large number of plants including citrus, coffee, guava, mango, persimmon, pear and quince. The hosts listed are mostly compiled from De Lotto (1965), Snowball (1969), Williams and Watson (1990), Ben-Dov (1993) and Qin and Gullan (1994, 1999). More host plants are listed by Zeck (1932) for New South Wales and Brimblecome (1956) for Queensland, Australia. In addition to the hosts listed,

the following are also host plants of *C. destructor*: *Alyxia ruscifolia, Bursaria spinosa, Conyza* sp., *Cussonia spicata, Dicliptera* sp., *Elaeodendron capensis, Maytenus heterophylla, Maesa* sp., *Maytenus* sp., *Pavetta revoluta* and *Pittosporum crassifolium*.

Primary hosts
Citrus, *Citrus aurantium* (sour orange), *Citrus maxima* (pummelo), *Citrus reticulata* (mandarin), *Citrus sinensis* (navel orange) (CAB 2004).

Secondary hosts
Acacia (wattles), *Actinidia deliciosa* (kiwi), *Azadirachta indica* (neem), *Camellia sinensis* (tea), *Coffea* (coffee), *Coffea arabica* (arabica coffee), *Coffea canephora* (robusta coffee), *Diospyros kaki* (oriental persimmon), *Dodonaea*, *Dodonaea viscosa* (switch sorrel), *Eremocitrus glauca* (Australian desert lime), *Gardenia*, *Hibiscus* (rosemallows), *Magnolia*, *Melia azedarach* (Chinaberry), *Persea americana* (avocado), *Plumeria* (frangipani), *Poncirus trifoliata* (Trifoliate orange), *Prunus armeniaca* (apricot), *Psidium guajava* (common guava), *Pyrus* (pears), *Rhus* (Sumach), *Schefflera*, *Schinus molle* (California peppertree), *Solanum* (nightshade), *Syzygium*, *Syzygium malaccense* (malay-apple), and *Theobroma cacao* (cocoa) (CABI 2004).

Known Distribution
Ceroplastes destructor is native to Africa and introduced to Australia, New Zealand and other South Pacific islands. Qin (2000) clarified some doubtful and unreliable distribution records of this species in the literature. A record of the pest in India (Avasthi and Shafee, 1986) is a misinterpretation of an earlier publication by Subba Rao (1965).

Potential Distribution within the US
It was recorded from Florida and Mexico (Ebeling, 1959; CIE, 1960); however, this was probably a misidentification of *Ceroplastes dugesii* (CAB, 2004).

Survey
Infestations of *C. destructor* on citrus and other hosts are easily detected because of their white wax cover. *Ceroplastes destructor* can be detected by examining and inspecting plants, especially shrubs or trees, for white wax covers, or for signs of sooty mold or sticky honeydew on leaves, branches and stems, or ants. To be certain of the presence of *C. destructor*, it is necessary to examine slide-mounted specimens under a microscope.

Key Diagnostics
Ceroplastes destructor was misidentified as *Ceroplastes ceriferus* in early literature, which is due to the similarity of the wax test of the two species. With the microscopic study of slide-mounted specimens, *C. destructor* can be morphologically distinguished from *C. ceriferus* by the absence of tubular ducts on the venter of the head in *C. destructor*. It also differs from other species of *Ceroplastes* by the possession of different-sized claw digitules (one slender and one broad) and a large and round group of spiracular setae (De Lotto, 1965; Williams and Watson, 1990; Qin and Gullan, 1994).

Ceroplastes japonicus

Scientific Name
Ceroplastes japonicus Green

Common Name(s)
Japanese wax scale, tortoise wax scale

Type of Pest
Scale insect

Taxonomic Position
Class: Insecta, **Order:** Hemiptera, **Family:** Coccidae

Reason for inclusion in manual
National pest list

Pest Description
Mature adult females of *C. japonicus* are greyish to pinkish-white, hemispherical and up to 4.0 mm in length and 3.5 mm in width (Fig. 1). They are covered by a thick layer of oily wax.

Authoritative identification involves detailed microscopic examination of newly matured adult females. Morphological descriptions and illustrations of the adult female are given by Pellizzari and Camporese (1994) and the immature female stages are described by Camporese and Pellizzari (1994). A key to the *Ceroplastes* species that occur in the Mediterranean is given by Pellizzari and Camporese (1994).

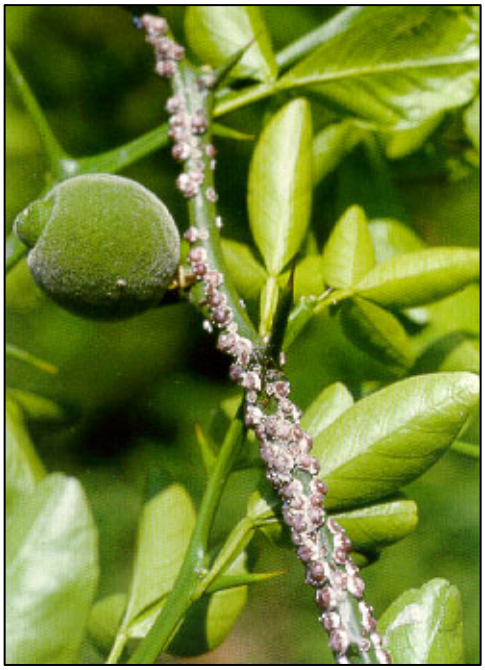


Figure 1: *C. japonicus* on ornamental plant. Photo courtesy of Regione del veneto

Pest Importance
Ceroplastes japonicus is a pest of ornamentals in towns and nurseries in northern and central Italy, but it is not yet recorded as a pest in southern Italy (Pellizzari and Camporese, 1994). It is also a pest of ornamental plants in urban environments in Japan, a major local pest of jujube trees in Zhejiang, China (Luo et al., 1994), a pest of citrus, mulberry, persimmon and fruit trees in western part of the Republic of Georgia (Borchsenius, 1957) and a pest of citrus and subtropical crops in southern Russia (Prokopenko and Mokrousova, 1981). In addition to the direct feeding damage, the honeydew secreted forms a substrate for the growth of black sooty molds, which screen light from the leaves and impair gas

exchange and photosynthesis. Sooty mold also reduces the market value of plants and produce (CABI, 2004).

Symptoms

Infestations of *C. japonicus* occur on foliage, stems and branches. This results in reduced vigor and the debilitation of the host plant. Heavy infestations may result in chlorotic spotting, the premature shedding of leaves, wilting and the dieback of stems. Honeydew deposited on the leaves and fruit serve as a medium for the growth of black sooty molds. The sooty mold results in a reduction of photosynthetic area and lowers the market value of ornamental plants and produce (CABI, 2004).

Known Hosts

Primary hosts

Camellia sinensis (tea), *Citrus*, *Citrus deliciosa* (mediterranean mandarin), *Citrus reticulata* (mandarin), *Diospyros kaki* (oriental persimmon), *Hedera helix* (ivy), *Ilex aquifolium* (Christmas holly), *Jasminum* (jasmine), *Laurus nobilis* (bay laurel), *Poncirus trifoliata* (Trifoliate orange), *Prunus* (stone fruit), and *Ziziphus jujuba* (common jujube) (CABI, 2004).

Secondary hosts

Acer japonicum, *Acer pseudoplatanus* (sycamore), *Camellia japonica* (Camellia), *Cleyera japonica*, *Cornus mas* (cornelian cherry), *Cycas revoluta*, *Cydonia vulgaris*, *Ehretia acuminata*, *Elaeagnus pungens*, *Elaeocarpus decipiens*, *Eriobotrya japonica* (loquat), *Euonymus japonicus*, *Fatsia japonica* (Japanese aralia), *Feijoa sellowiana* (Feijoa fruit), *Ilex integra*, *Magnolia grandiflora* (Southern magnolia), *Malus* (ornamental species apple), *Mangifera indica* (mango), *Morus* (mulberrytree), *Myrtus communis* (myrtle), *Nerium oleander* (oleander), *Persea thunbergii*, *Pittosporum tobira* (Japanese pittosporum), *Platanus orientalis* (plane), *Podocarpus nagi*, *Prunus avium* (sweet cherry), *Prunus laurocerasus* (cherry laurel), *Prunus mume* (Japanese apricot tree), *Prunus persica* (peach), *Prunus yedoensis*, *Pyracantha coccinea* (Scarlet firethorn), *Pyrus communis* (European pear), *Pyrus pyrifolia* (Japanese pear), *Salix* (willow), *Trachelospermum asiaticum*, and *Ulmus minor* (European field elm) (CABI, 2004).

Known Distribution

According to Borchsenius (1949), *C. japonicus* originates in eastern Asia and was accidentally introduced into the Republic of Georgia, southern Russia, Italy and France. It was first detected in Italy in 1983 and has since spread throughout the country, as it continues to increase its range (Camporese and Pellizzari, 1994; Pellizzari and Camporese, 1994). It also appears to be spreading in the Republic of Georgia (Dekanoidze, 1971).

Green (1921), Boratynski and Williams (1964) and Ben-Dov (1993) record *C. japonicus* in the United Kingdom, but it is currently not established in the UK.

Potential Distribution within the US

Very little information is available on the potential distribution of *C. japonicus* within the US. The pest risk potential for *C. japonicus* is classified as high according to Cave and Sutker (2003).

Survey

Adult female *C. japonicus* are conspicuous as they are covered by a thick layer of greyish to pinkish-white, oily wax that contrasts in color with the host plant. Heavy infestations are conspicuous and the foliage, fruit and stems of the plant become covered in sticky honeydew, serving as a medium for the growth of black sooty molds. *Ceroplastes japonicus* is polyphagous, attacking more than 100 plant species belonging to 40 genera placed in 24 families, including many crop and ornamental plants (Ben-Dov, 1993; Pellizzari and Camporese, 1994). The most common host plants are Citrus, persimmon, holly and ivy. In the Republic of Georgia, it is also common on mulberry and fruit trees, and in Italy, on bay laurel and maple (Pellizzari and Camporese, 1994).

Key Diagnostics

Ceroplastes japonicus should be distinguished from the closely related *C. floridensis*, which occurs worldwide in tropical and subtropical regions. The main characters used to distinguish the two species are the number and different arrangements of stigmatic setae along the body margin. In *C. japonicus* the stigmatic setae form an uninterrupted row between the anterior and posterior stigmatic clefts, whereas, in *C. floridensis*, they are interrupted with 7 to 12 bristle-shaped marginal setae. *Ceroplastes japonicus* has an average of 111 stigmatic setae on each side of the body compared with an average of 60 stigmatic setae in *C. floridensis* (Pellizzari and Camporese, 1994).

Paratachardina lobata lobata

Scientific Name
Paratachardina lobata lobata Chamberlin

Common Name(s)
Lobate lac scale

Type of Pest
Scale insect

Taxonomic Position
Class: Insecta, **Order:** Hemiptera, **Family:** Kerridae

Reason for inclusion in manual
National pest list

Pest Description
The mature females of *P. lobata lobata* are about 1.5 to 2 mm long, and about the same width (Fig. 1). The body has two pairs of prominent lobes. To the practiced eye, this scale insect's “X-shaped” appearance is discernable, even without magnification. The testa is extremely hard and brittle, glossy and of a dark reddish brown color, which often appears dull and black due to a coating of sooty-mold. The first instars (crawlers) are elongate-oval, deep red, and about 0.4 mm long (Fig. 2). The characteristic lobate pattern develops in the second instar. The second instar female presumably molts to the adult female as in other scale insects. Males of this species have not been observed in Florida.

Figure 1. Mature females of *P. lobata lobata*. Photo courtesy of F.W. Howard.

The resinous scale covering is light to dark reddish brown. Old individuals will frequently appear black because of the sooty molds. The shape is globose with four lobes; young individuals generally appear more lobed than mature adults. The case size is about 1.5 mm long and 1.0 mm wide, but individuals in close proximity will frequently coalesce forming masses of individuals. Young individuals often appear like a "fat bow tie." The resinous case conforms approximately to the shape of the insect inside. Exactly how the insect grows larger within such a rigid case is not understood entirely.

Pest Importance

Paratachardina lobata lobata is potentially one of the most devastating pests of trees and shrubs in Florida history. This scale insect has been found on woody dicotyledonous plants, on one coniferous species, the southern red cedar, *Juniperus silicicola*, and the palm, *Phoenix roebelenii*. As of October 2002, more than 120 species in 44 families of woody plants, many of which are economically important, have been determined to be hosts of *P. lobata lobata* in Florida, including 39 plant species native to Florida. Most of the exotic host plants are grown as ornamental shrubs or trees, or as fruit trees. Some of these are extremely important in the urban landscape as shade trees, specimen trees, or hedges.

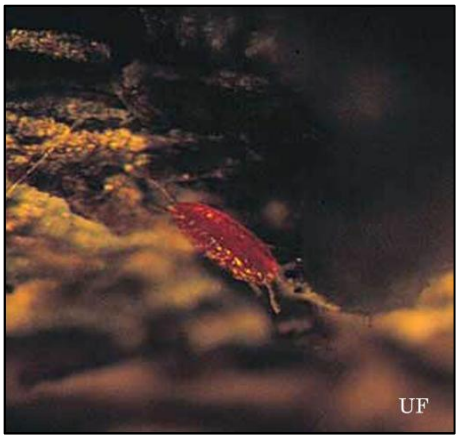


Figure 2: First instar of *P. lobata lobata*. Photo courtesy of F.W. Howard.

Paratachardina lobata lobata is known to reach damaging levels on mango, carambola, cinnamon, avocado, hibiscus, starfruit, pomegranate, crab apple, gardenia, sour orange, grapefruit, kumquat, lychee, and sapodilla, which are raised in the continental US, Hawaii, Puerto Rico, the Virgin Islands, and the Pacific Islands of the US (Mayerdirk, 2003). Countries that import nursey stock from Florida could impose restrictions due to possible infestations and potential transport of lobate lac scale on nursery stock.

Symptoms

Paratachardina lobata lobata has been found on woody dicotyledonous plants. It infests the woody portions of twigs and small branches and, less frequently, the main stems that are usually < 2 cm in diameter, but not > 2 cm in diameter. It has not been observed on foliage. On highly susceptible hosts, the scale insects are crowded and form a contiguous mass that appears to be a dark lumpy crust. On wax-myrtle (*Myrica cerifera*), a highly susceptible host, up to 42 mature females have been counted per 1 cm segment of twig (Fig. 3). Sooty mold (Fig. 4) covers the branches with insects, occurring in patches on the foliage. Dense infestations are associated with branch dieback of some plant species and, in severe cases, highly infested shrubs and small trees have died. Wax myrtle is especially prone to become heavily infested and die from the effects of lobate lac scale. Some plant species appear to tolerate dense infestations, but this may be illusory, as the long-term effects of such infestations are not yet known.

Figure 3. Wax-myrtle branch infested with lobate lac scale. Photo Courtesy of F.W. Howard.

Known Hosts

This scale insect has been found on woody dicotyledonous plants, on one coniferous species, the southern red cedar, *Juniperus silicicola*, and on the palm, *Phoenix roebelenii*. More than 300 species in more than 50 families of woody plants have been determined to be hosts of *P. lobata lobata* in Florida, including 39 plant species native to Florida. Economically important hosts include avocados, cinnamon, pears, apple, *Citrus spp.* (including grapefruit and pomelo), green coffee, legumes, and mangoes.

Figure 4. Sooty mold on mango leaves, an indirect result of infestation by lobate lac scale. Photo courtesy of F.W. Howard.

Some plant families, notably Fabaceae, Myrtaceae, and Moraceae, are especially well-represented by species that serve as hosts, which may be related to their abundance in the landscape or other biases. Plants at different sites have been exposed to infestations for different periods; the levels of infestation are highly variable. Differences in susceptibility have not been experimentally determined; however, certain species appear to be highly susceptible, including certain natives, e.g., wax-myrtle, cocoplum (*Chrysobalanus icaco*), buttonwood (*Conocarpus erectus*), strangler-fig (*Ficus aurea*), myrsine (*Myrsine guianensis*), red bay (*Persea borbonia*), and wild-coffee (*Psychotria nervosa*); popular exotic ornamental plants, e.g., black-olive (*Bucida buceras*), Indian laurel (*Ficus microcarpa*), Benjamin fig (*F. benjamina*); and fruit trees, e.g., lychee (*Litchi chinensis*), mango (*Mangifera indica*), and star-fruit (*Averrhoa carambola*).

Known Distribution

The species belongs to the lac scale family, Kerriidae, the best-known species of which is the true lac scale insect, *Kerria lacca lacca* (Kerr). The testa of the true lac scale insect has been utilized for centuries for making shellac and similar products. Most species of the family, including *P. lobata*, do not produce any material of known commercial value. The specific scientific name, lobata, refers to the four prominent projections, or lobes, of this scale, and the vernacular name 'lobate lac scale' may be used for this species.

Of the 28 families of Coccoidae recognized by Miller and Ben-Dov (2002), 13 are represented by species native to Florida (Aclerdidae, Asterolecaniidae, Diaspididae, Cerococcidae, Coccidae, Conchaspidae, Dactylopiidae, Eriococcidae, Kermesidae, Lecanodiaspididae, Margarodidae, Ortheziidae, and Pseudococcidae). No species of Kerriidae is native to Florida and adjacent land areas. The Kerriidae is confined to the tropics, with a minority of species found in low latitude desert areas. Of the 87 described species, 64 are distributed in the eastern hemisphere. Of the species native to the western hemisphere, 13 are reported from South America, six from Mexico (two of which are also reported in the southwestern U.S.), three reported in the southwestern U.S., and one from Jamaica (Ben-Dove, 2002).

The lobate lac scale, native to India and Sri Lanka, was found for the first time in Florida in August 1999 by personnel of the Florida Department of Agriculture and Consumer Services, Division of Plant Industry (DPI) (Hamon, 2001). The identification of the species by Avas Hamon of DPI was confirmed by D. R. Miller of the Systematic Entomology Laboratory, U.S. Department of Agriculture, Beltsville, MD. This first record was on a hibiscus (*Hibiscus rosa-sinensis*) in the town of Davie (Broward County). The plant was destroyed by DPI personnel. Plants in the vicinity of this infested hibiscus were inspected without finding *P. lobata.* The species was found again in 2000 on a Benjamin fig (*Ficus benjamina*) in Davie, on cocoplum (*Chrysobalanus icaco*) in Weston (Broward County), and on cocoplum at two sites in Miami (Miami-Dade County). In 2001, the scale insect species was found on 11 sites in Broward County and six sites in Miami-Dade County. In December 2001, DPI inspectors found *P. lobata* in Lake Worth (Palm Beach County). As of October 2002, *P. lobata* has been recorded from sites from Lake Worth on the north to Homestead (Miami-Dade County), a distance of 128 km, and from the coast to 28 km inland. In 1992, specimens of scale insects submitted to DPI from the Bahamas had been identified as *P. lobata.*

Potential Distribution within the US

The potential for further spread of this scale insect in the western hemisphere is especially high for warm areas into which there is significant movement of living plants from Florida, e.g., Puerto Rico and other localities of the Caribbean Region, California, and Hawaii.

The invasion of natural areas is of paramount concern. A cursory examination in several tropical hardwood hammocks in Broward County revealed that there were heavy infestations on diverse species over large areas. The presence of heavy infestations 28 km inland, i.e., virtually at the eastern edge of the Everglades, implies that vegetation in this vast natural area is threatened. Most of the native host plants of *P. lobata* identified in Florida are also distributed in the Caribbean Region. If the insect were to be introduced into Puerto Rico or other Caribbean countries, natural areas they would likewise be threatened.

Survey

Paratachardina lobata lobata has been found on woody dicotyledonous plants. It infests the woody portions of twigs and small branches and, less frequently, the main stems usually < 2 cm in diameter, but not branches or main stems > 2 cm in diameter. It has not been observed on foliage. On highly susceptible hosts, the scale insects are crowded, forming a contiguous mass that appears as a dark, lumpy crust. Sooty mold becomes obvious and covers the branches, scale, and leaves. Where sooty mold is thick on the leaf surface, little photosynthesis takes place. Die back of severely infested branches will occur, and, if left untreated, plants will die.

The scale infests woody portions of twigs under one-inch diameter and small branches. It attaches to the branch and sucks the plant sap, causing the host plant to starve from lack of food and water. Woody portions of citrus trees should be visually examined for the presence of mature lobate lac scale females and sooty mold. Each plant should be scanned for about five minutes. Particular attention should be placed on woody areas of the plants with sooty mold.

Key Diagnostics

If a scale insect is found with the “fat bow tie" shape, samples should be sent to the appropriate diagnositic laboratory for confirmation.

Pulvinaria polygonata

Scientific Name:
Pulvinaria polygonata Cockerell

Synonyms
Chloropulvinaria polygonata

Common Name
Cottony Citrus Scale

Type of Pest
Scale Insect

Taxonomic Position
Class: Insecta, **Order:** Hemiptera, **Family:** Coccidae

Reason for inclusion in manual
National pest list

Pest Description
There is limited information available on this pest. No descriptions or images of the scale insect were found. The scale insect is generally found on mango trees, in addition to the host plants: citrus, palm, banana, and sugarcane. They infest the tender parts of the plants and trees, feeding on plant sap. Very little is known about their life and seasonal history.

Pest Importance
Until recently, there have been few reports of this scale insect being a pest in citrus or mangos. *Pulvinaria polygonata* poses a serious threat to mango industry of western Uttar Pradesh, India. In this area, *Pulvinaria polygonata* is the predominant species; however, mixed populations with *Aspidiotus destructor, Ceroplastes* spp. and *Rastococus* spp. are fairly common. The nymphs and adult scales suck the sap of the leaves and other tender parts, reducing the plant's vigor. They also secrete honeydew, which encourages the development of sooty mold on leaves and other tender parts of the mango plant. In case of severe scale infestation, growth and fruit bearing capacity of the tree is adversely affected.

Symptoms
Scale insects can be very cryptic and difficult to locate in the field. The presence of sooty mold can be used as a preliminary indication of pest presence. Sooty mold often covers the upper leaf surfaces, causing the leaf to turn black in color. Scale insects, particularly the armored species, are very destructive and persistent mango pests. They feed by sucking the plant sap causing affected parts to dry up and shrivel. Feeding on branches/twigs will result in the abnormal growth of tissues that produce gall-like protuberances. Damage

symptoms become more prevalent during the summer months, as affected foliage loses vigor, ceases in growth, and eventually dies.

Known Hosts
Citrus spp., Citrus deliciosa (Mediterranean mandarin), *Citrus reticulata* (mandarin) *Mangifera indica* (mango)

Known Distribution
Australia, India, and the Philippines

Potential Distribution in the US
Presumably, *P. polygonata* could become established in locations in the US where citrus and mangoes are grown, which are primarily tropical and subtropical growing environments.

Survey
No specific survey methods are identified in the available literature. Surveys in citrus and mangoes should use damage symptoms to identify affected foliage.

Key Diagnostics
None available

Unaspis yanonensis

Scientific Name
Unaspis yanonensis Kuwana

Common Name(s)
Japanese citrus scale, Oriental citrus scale, citrus snow scale, arrowhead scale.

Type of Pest
Scale insect

Taxonomic Position
Class: Insecta, **Order:** Homoptera, **Family:** Diaspididae

Reason for inclusion in manual
National pest list

Pest Description
Adult female scale covers are oyster-shell shaped, 2.5 to 3.6 mm, blackish-brown with a paler margin (CABI, 2004). Exuviae terminal are brownish-yellow. Immature male scale covers are elongate, 1.3 to 1.6 mm, felted white, with two or three longitudinal ridges (see USDA, 1984 - which also gives full diagnostic characters) (Fig. 1)

Pest Importance
Unaspis yanonensis feeds almost exclusively on *Citrus spp*. In Japan, it is found on all types of citrus except on the Japanese hybrids known as Natsudaidi and on Citrus junos (Ohkubo, 1980). It affects fruits, leaves, stems—the whole plant—and can cause serious damage to orchards due to leaf drop and rapid dieback.

Figure 1. Arrowhead scales on branches and leaves. Photo courtesy of http://perso.wanadoo.fr/scanice/cochenille.htm)

Fruits, leaves and small branches are attacked, whereas large branches and trunks are not. Only the second and third generations are found on fruits (Ohkubo, 1980). Attacked fruits lose their commercial value because of the feeding punctures of the pest. Relatively low numbers of scales can cause damage. Leaves and branches begin to die back at a density of 1.1 females per leaf (Ohkubo, 1980), while, in the spring, a density of 8 females per leaf is likely to lead to complete dieback of the tree within the year (Ohgushi and Nishino, 1968). The cause of the dieback is not yet understood, but it has been suggested that the scale may inject toxic saliva into the tissues.

Symptoms

Attacked plants show inhibited growth, yellow blotches and necrosis of leaves, leaf fall, shortened or dead branches, and small deformed fruits. Large masses of male white scales may be seen on twigs with darker curved female scales. In cases of severe attacks, tree mortality has been observed (CABI, 2004).

Known Hosts

Unaspis yanonensis feeds almost exclusively on *Citrus spp*. Primary hosts include *Citrus deliciosa* (Mediterranean mandarin), *Citrus limon* (lemon), *Citrus reticulata* (mandarin), *Citrus sinensis* (navel orange), *Citrus unshiu* (satsuma), and *Citrus x paradisi* (grapefruit). It has been reported on *Damnacanthus* in Rubiaceae, *Fortunella* (kumquat) and *Poncirus* in Rutaceae (USDA, 1984).

Known Distribution

Unaspis yanonensis originates from Southeast Asia, and has been accidentally introduced into limited areas in southern France and northern Italy (Liguria). *Unaspis citri* is present in China, Japan, Korea, France, Italy, and Fiji (CABI, 2004).

Potential Distribution within the US

This predominately Asian species prefers the warm temperate Mediterranean and tropical climates; citrus is its host species. There is potential for the scale insect, *U. yanonensis*, to become established in particular areas of the US where citrus production and warm temperatures co-exist.

Survey

In Japan, plants damaged by this scale typically have leaves, withered green twigs, and whole branches that are dead. Feeding by this scale seems to cause inordinate amounts of damage; very lightly infested leaves will wilt and die.

Heavily infested trees are conspicuous and easily recognized by large masses of white male scale covers on the twigs, leaves, and fruit. The small size, dark color, and sessile nature of the female scales make them difficult to detect unless present in large numbers. On citrus fruit, the female scales can be confused with the common *Lepidosaphes spp*. or easily overlooked as dirt particles.

Individual leaves, stems, and fruit should be examined for various stages of the scale insect. If scale insects are present, material should be forwarded to a laboratory for microscopic examination and positive identification. When forwarding material for

identification, scales should be attached to the host material. Adult females are important for the identification and speciation, should be included in the material if possible. Because the pathogen is exotic, be sure the specimens are dead. The easiest way to accomplish this is to freeze the leaf, twig, and fruit for up to 24 hours (USDA, 1984).

Key Diagnostics

Identification of armored scales is performed by studying the shape, dimension, and color of the cover. For this reason, the infested parts of the plant have to be examined under a stereomicroscope. For an exact identification to species, the body of adult females should be studied, as there are no adequate keys for the separation of species based on nymphs or adult males. Covers of specimens are moved with a needle to collect scale bodies. Specimens removed from under the cover have to be prepared for morphological study under the microscope.

The arrowhead scale is similar to the citrus snow scale, *Unaspis citri*, but can be distinguished from it by microscopic examination of the adult females. *Unaspis citri* is present in California, Florida, Georgia, and Louisianna. Adult female *U. citri* have relatively few pygidial dorsal ducts, do not have marked divisions between the thoracic segments, and have subjacent median lobes. Adult female *U. yanonensis* have numerous pygidial dorsal ducts, usually marked divisions between the thoracic segments and distinct median lobes (USDA, 1984; CABI, 2004).

Thrips

Scirtothrips citri

Scientific Name
Scirtothrips citri Moulton

Synonyms
Euthrips citri, *Scirtothrips clivicola*

Common Name(s)
California thrips, citrus thrips

Type of Pest
Thrips

Taxonomic Position
Class: Insecta, **Order:** Thysanoptera, **Family:** Thripidae

Reason for inclusion in manual
Western Region pest list

Pest Description
Scirtothrips citri was originally described as *Euthrips citri* by Moulton in 1909. This species was transferred to the genus *Scirtothrips* in 1914. This genus includes more than 60 species worldwide, many of which can cause damage to important crops.

Figure 2. Immature larvae of *Scirtothrips citri*. Photo courtesy of Jack Kelly Clark. Copyright © 2005 Regents of Unversity of California

Figure 1. Adult of *Scirtothrips citri*. Photo courtesy of Jack Kelly Clark. Copyright © 2005 Regents of Unversity of California

Adult citrus thrips are small, orange-yellow insects with fringed wings. Males are similar to females (Fig. 1), but smaller. The females measure 0.6 to 0.88 mm. They can lay about 25 eggs (0.2 mm in size) under the cuticle of new leaves, flowers, green twigs, or young fruit. Eggs usually hatch in 5 to 15 days at temperatures above 20°C. The first and second instars, which last between 4 to 10 days, are active larval stages. First-instar larvae are very small, whereas second-instar larvae are about the size of adults, spindle-shaped, and wingless (Fig. 2). They feed actively on tender tissue, especially under the sepals of young fruit. The third (prepupa) and fourth-instars (pupa) are pupation stages, last

3 to 5 days, and do not feed. Pupation mainly occurs on the trunks of trees, but 20 to 30% of larvae pupate in the soil (Grout et al., 1986). Other studies have indicated that a higher proportion of the population pupates in the leaf litter and upper soil layers (Schweizer and Morse, 1996). Temperature can affect the rate of development. The development time can range from about 12 days at 35ºC to 35 days at 18ºC (Tanigoshi and Nishio-Wong, 1982). When adults emerge, they move actively around the tree foliage. If the weather is favorable, 8 to 10 generations per year may be produced. Under warm conditions, an adult may live as long as 25 to 35 days (Kerns, et al., 2004).

Biology and Ecology
In California, *S. citri* overwinters at the egg stage. Overwintering eggs are laid in the last flush growth of the season. Eggs hatch as nymphs in February and March. These nymphs pupate and emerge as adults in March and early April, completing the first generation. Adult females appearing in March lay the second generation of eggs. Nymphs hatching from these eggs in late March and April are the most damaging and responsible for the scarring of citrus fruit (Rhodes, et al., 1989a). A similar pattern was found in Arizona, but it was not uncommon to find citrus thrips year-round during warm periods (Rethwisch, et al., 1998). Economic damage is mainly caused by larvae developing and feeding on the very young citrus fruits (Wiesenborn and Morse, 1986).

In California, citrus thrips have several natural enemies, including the predaceous mites *Amblyseius degenerans and Euseius tularensis*, spiders, lacewings, dustywing, and minute pirate bugs. Under controlled conditions, only *A. degenerans* had a beneficial effect on plant growth by depressing the level of thrips populations (Grafton-Cardwell, et al., 1999).

Pest Importance

Although damage by citrus thrips is entirely cosmetic, *S. citri* is considered to be the most economically damaging pest of citrus in Arizona and California. Control of citrus thrips has cost Arizona growers as much as $618/ha in pesticides, and packing lines had to slow down to remove the scarred fruit (Bates, 1991). In California, the estimated average cost was $116/ha (Morse 1995). The scarred fruit are processed rather than sent to fresh market, which results in an economic loss. Heavily scarred fruit may also rapidly lose weight (Arpaia and Morse, 1991).

Figure 3. Symptoms of citrus thrips damage in orange fruit. Photo Courtesy of D. Kerns, University of Arizona.

Symptoms

The most typical symptom of damage by *S. citri* on citrus fruit is the grayish or silvery ring of corky tissue on the skin around the fruit apex (Fig. 3). This damage is mainly caused by the feeding of the second-instar larvae beneath the shelter provided by the calyx remnants. As the fruit grows, damaged rind tissue moves outward from beneath the sepals and a conspicuous ring of scarred tissue is evident. Damage can also be present where two fruits touch each other. These areas provide thrips with protection, and larvae may aggregate and feed causing lateral corky blemishes. Fruits are more susceptible to scarring shortly after petal fall and until they have reached about 3.7 cm in diameter. Thrips damage is higher on fruit located on the outside the canopy

(Olendorf et al., 1994). Feeding of *S. citri* on new flush may result in distorted and thickened leaves with gray streaks usually parallel to the midvein (Kerns et al., 2004).

Known Hosts

The primary hosts are *Citrus spp. Scirtothrips citri* is believed to be native to California, Arizona and northwestern Mexico. Its primary natural host-plant appears to be *Rhus laurina* (Anacardiaceae) (Morse, 1995). Since citrus is an introduced crop in California, the host-shift made by *S. citri* is particularly remarkable.

Adults of *S. citri* have been collected from a wide range of plants (56 species), including some of the most economically important species: avocado, alfalfa, mango, rose, grape, peach, olive, cotton and others (Morse, 1995). Despite this, it is generally assumed that Californian *S. citri* lives on citrus, and has only a limited immigration into the crop from other plants and surrounding areas.

Known Distribution

Scirtothrips citri is present in citrus groves in California and Arizona (Morse, 1995, Kerns, et al., 2004). It is mainly established in muscadines grapes in northern Florida (Flowers, 1989), and it may be present in Mexico. *Scirtothrips citri* is not yet found in Texas. Published references from other countries have not been confirmed.

Potential Distribution in the US

Present in California, Arizona, and Florida.

Survey

Citrus thrips are usually first detected through the damage they cause to young foliage. The detection of low-level populations involves careful examination of plant parts for the presence of the small pale adults and minute larvae.

The best method for surveying citrus thrips is by the use of a beating tray. New flush may be sampled from the circumference of the tree in a range from knee to neck height, between 10:00 and 14:00 h (Rhodes and Morse, 1989).

Several methods have been reported to monitor citrus thrips on navel oranges (Rhodes and Morse, 1989b). These methods may vary according to the life stage that is intended to be monitored. Sticky traps may be used to monitor late second instars falling from the trees to pupate. In this case, one sticky trap is placed under each tree with the center of the trap about 30 cm from the tree trunk and changed twice per week. Five non-adjacent trees can be sampled. Pupation papers are useful to monitor late second instar moving towards the tree interior. Blue paper towels are folded twice and attached with small binder clips to horizontal twigs about 30 cm inside the canopy. Each paper is placed on branches with new foliage, at 1.4 m above the ground. Samples are taken from the SE and NE quadrants of 20 non-adjacent trees, and counts can be made twice per week. Sticky glossy-yellow polyvinyl cards (7.6 x 12.7 cm) can detect adult thrips. These cards are suspended at 1.5 m above the ground and on the outside of the canopy. Cards can be placed in the SE quadrant of five non-adjacent trees and evaluated twice per week. Populations of first and second instars and adults can be determined on fruit. Twenty fruit can be sampled at random from

the circumference of five non-adjacent trees, or five fruit from the N and S sides of each of 20 non-adjacent trees.

In the case of lemons, populations of adults and immature citrus thrips on fruit can be estimated by sampling twenty fruit per tree (five from each side) from 10 trees (Kerns and Tellez, 1998), or two fruit from each of the four sides of 50 trees (Kerns, et al., 1997). A beat-pan method can be used to determine adult and immature thrips in flush growth. The flush is gently tapped against a hardware cloth covering a black cake pan. One beat-pan sample from four sides of 50 trees can be examined in each grove (Kerns, et al., 1997). Lemons are most susceptible to damage by *S. citri* from petal fall until the fruit reaches 2.54 cm (1 inch) in diameter (Kerns and Tellez, 1998).

Key Diagnostics

Scirtothrips citri is similar to the South African citrus thrips; *Scirtothrips aurantii* and *S. dorsalis* are found on many different plants across the Old World tropics to Australia. Both of these species have a dark area medially on the abdominal tergites, transverse dark lines on the tergites and sternites, and a pair of setae close together between the posterior pair of ocelli on the head (EPPO, 2004).

Other species of thrips can occur in citrus fields. In Arizona, *Frankliniella occidentalis,* the western flower thrips, is very common during its blooming period and on small fruit and flush growth; they do not cause any damage (Kerns, et al., 2004). Adults of *S. citri* have oval shaped abdomens, are active crawlers and flyers, light orange-yellow in color, and 0.5 to 0.88 mm in size; whereas, those of *F. occidentalis* have long cigar-shaped abdomens, are relatively sluggish, straw- or dark brown in color, and 1.0 mm in size.

The fruit scarring brochure published by the University of California may help to differentiate citrus thrips scarring from other scarring caused by biotic or abiotic agents (http://anrcatalog.ucdavis.edu/InOrder/Shop/ItemDetails.asp?ItemNo=8090).

Scirtothrips dorsalis

Scientific Name
Scirtothrips dorsalis Hood

Synonyms:
Anaphothrips andreae, Heliothrips minutissimus, Neophysopus fragariae, Scirtothrips andreae, S. fragariae, S. minutissimus, S. padmae

Common Name(s)
Castor thrips, chilli thrips, yellow tea thrips

Type of Pest
Thrips

Taxonomic Position
Class: Insecta, **Order:** Thysanoptera, **Family:** Thripidae

Reason for inclusion in manual
National pest list

Pest Description
Scirtothrips dorsalis is a widespread pest described as a new species by Hood in 1919. *Scirtothrips dorsalis* is very small (0.8 to 1.0 mm) and pale yellow in color (Fig. 1). It can be found feeding on leaves, flowers, and calices of fruit on a wide variety of host crops. The CAPS Pest Risk Analysis (Venette & Davis, 2004) provides a thorough review of its host plants known from scientific literature. Recent field surveys in the Caribbean have shown that *S. dorsalis* was found on cucumbers, cantaloupe, watermelon, pumpkin and squash, hot peppers, tomato, eggplant, okra, and beans (Ciomperlik & Seal, 2004).

Figure 1. *Scirtothrips dorsalis* adults. Courtesy of T. Skarlinsky (USDA APHIS PPQ)

Egg: Typically oval, whitish to yellowish, narrow anteriorly, incubation period 4 to 6 days.

Larva: First instar transparent; body short, legs longer; antennae short, swollen; mouth cone bent and short; and antennae seven-segmented and cylindrical. Sclerotization not distinct, head and thorax, reticulate. Second instar antennae longer, cylindrical, seven-segmented; mouth cone longer; maxillary palpi three-segmented; body setae longer than the first instar; head and thorax reticulate with sclerotization of head.

Pre-pupa: Yellowish; antennae swollen, short, with distinct segmentation; two pairs of external wing buds on each meso- and meta-thorax.

Pupae: Dark yellow with eyes and ocelli bearing red pigmentation; wing buds are elongate; antennae short and reflected over the head; female pupae with larger pointed abdomen that of male smaller, with blunt abdomen.

Adult: Almost white on emergence, turning yellowish subsequently (Fig. 1).

Pest Importance

On chilies, *S. dorsalis* causes 'leaf curl disease.' The pest occurs in such large numbers that the young leaves shrivel. Heavy infestation of the tender shoots, buds and flowers causes the leaves to curl, when shed fresh buds become brittle, subsequently dropping off. During bad seasons, 25 to 55% of the total yield is lost (Ramakrishna Ayyar, 1932; Ramakrishna Ayyar and Subbiah, 1935). Ramakrishna Ayyar (1932) recorded this species as a major pest in south India causing the 'Murda disease', or dying back, of the young seedlings.

Scirtothrips dorsalis is also a vector of tomato spotted wilt virus (TSWV) which causes bud necrosis disease (BND), a disease of peanut in India (Amin et al., 1980; Mound and Palmer, 1981; Ananthakrishnan, 1993)

Symptoms

Deformed leaves, leaf senescence, stunted plants, loss of vigor, and yield loss are common symptoms on all hosts. Being a serious pest on castor, *S. dorsalis* infests shoots, leaves, flowers and young fruits. The growing tip of the plant, the young leaves in particular, and the axillary leaf branches are the main targets of attack. The infested plant parts turn brown to black and, in extreme cases, there is total deformation and defoliation. Although infestation occurs throughout the year, it peaks during the drier months.

Figure 2. Leaf curling symptom. Photo courtesy of CABI, 2004.

On chillies, *S. dorsalis* causes 'leaf curl disease.' Heavy infestation of the tender shoots, buds and flowers cause the leaves to curl (Fig. 2), when shed, fresh buds become brittle, subsequently dropping off. This species causes damage to the soft part of the plant, particularly the shoots and leaves. Damage ranges from the slight disruption of the tissues to total deformation and disruption. The damage is due to continuous sucking of the cell sap, which leads to necrosis of the cell tissues. Eggs are also laid inside the soft tissues and the larvae leave large circular holes causing the deformation of plant parts; as a result, the plants may remain stunted due to the defoliation and deformation of the leaves.

Heavy infestation of cotton plants at the early seedling stage, in particular the cotyledons and other young leaves, result in brittle leaves and premature leaf drop. *Scirtothrips dorsalis* is also evident in mixed cropping of cotton, chillies and onion. Incidence and infestation of this species in different ecological conditions in cotton fields is also known (Ananthakrishnan, 1969, 1971, 1984).

Known Hosts

Scirtothrips dorsalis is known as a pest of many crops, including *Actinidia chinensis*, *Anacardium occidentale* (cashew nut), *Arachis*, *Capsicum* (chili and hot peppers), *Citrus*, cotton (*Gossypium hirsutum*), *Fragaria*, grapevine (*Vitis vinifera*), *Hevea brasiliensis*, *Hydrangea*, *Lycopersicon esculentum* (tomato), *Mangifera* (mango), *Nelumbo*, *Nicotiana tabacum* (tobacco), onions (*Allium cepa*), *Ricinus* (castor bean), *Rosa*, tamarinds (*Tamarindus indica*) and tea (*Camellia sinensis*). It is only cited as a significant pest of Citrus in Japan and Taiwan.

Known Distribution

Scirtothrips dorsalis is distributed throughout Bangladesh, Brunei, Darussalam, Hong Kong, Hainan, Guandong, Taiwan, Zehjiang, India, Indonesia, Israel, Japan, Republic of Korea, Malaysia, Myanmar, Pakistan, Philippines, Sri Lanka, Thailand, Africa, Hawaii, Australia, Papua New Guinea, and the Solomon Islands. It has recently been reported as an invasive species in Saint Lucia, Saint Vincent, Trinidad, and Tobago.

Potential Distribution in the US

Venette and Davis (2004) indicate that the potential geographic distribution of *S. dorsalis* in North America extends from southern Florida to north of the Canadian boundary, as well as to Puerto Rico and the Caribbean.

Survey

Scirtothrips dorsalis is found on the leaves, flowers, and fruits of the hosts listed in this document. The plant foliage is sampled at random and secured in a re-sealable zip-lock bag for processing in the laboratory. Foliage is washed in 70% ethanol to remove the adults and immatures. The alcohol and thrips samples are screened through a mesh diameter of 0.5 mm or less and observed under a stereomicroscope. Species level identifications should be made by a qualified taxonomist.

Key Diagnostics

Members of the genus *Scirtothrips* are readily distinguished from all other Thripidae by the following characteristics: surface of pronotum covered with many closely spaced transverse striae; abdominal tergites laterally with numerous parallel rows of tiny microtrichia; sternites with marginal setae arising at posterior margin; metanotum with median pair of setae arising near anterior margin. A similar species is *Drepanothrips reuteri*, a native European pest of grapevine; however, it has 6-segmented antennae instead of 8-segmented (the 3 terminal segments being fused).

Adult characters: abdominal tergites with median dark patch, tergites and sternites with dark antecostal ridge; ocellar setae pair III situated between posterior ocelli; 2 pairs of median post-ocular setae present; pronotum with four pairs of posteromarginal setae, major setae 25 to 30 µm long; metanotum medially with elongate recticles or striations, arcuate in anterior third, median setae not at anterior margin; forewings with fore marginal setae, second vein with two setae, cilia straight; tergal microtrichial fields with 3 discal setae, VIII and IX with microtrichia medially; sternites with numerous microtrichia, more than 2 complete rows medially; male without drepanae on tergite IX (Palmer and Mound, 1983).

Whiteflies

Aleurocanthus spiniferus

Scientific Name
Aleurocanthus spiniferus Quaintance

Synonyms:
Aleurocanthus citricolus, A. rosae, A. spiniferus var. intermedia, Aleurodes citricola, Aleurodes spinifera

Common Name(s)
Orange spiny whitefly, citrus mealywing

Type of Pest
Whitefly

Taxonomic Position
Class: Insecta, **Order**: Hemiptera, **Family:** Aleyrodidae

Reason for inclusion in manual
National pest list

Pest Description
Eggs: The following details are based on Smith et al. (1992). Eggs elongate to become oval in shape (0.2 mm long). Eggs are yellow when first laid and then darken to charcoal grey or black (Fig. 1); each is attached to the leaf by a short pedicel.

Larvae: The six-legged, dusky, elongated first-instar larvae (0.3 x 0.15 mm) have two long and several shorter, slender dorsal glandular spines. All subsequent immature stages are sessile, have non-functional leg stubs and possess numerous, dark dorsal spines on which a stack of exuviae of earlier instars may occur. The second instar (0.4 x 0.2 mm) is a dark brown to charcoal convex disc with yellow markings, while the third instar (0.87 x 0.74 mm) is usually black with a rounded, greenish spot on the anterior part of the abdomen and obvious dorsal spines. In the fourth immature stage or 'pupa', females are larger (1.25 mm long) than males (1 mm long). This stage is black, has numerous dorsal spines and is often surrounded by a white fringe of waxy secretion. This is the stage required for identification purposes.

Figure 1. *A. spiniferus* eggs and nymphs. Nymphal instars 1, 2 and 4 (pupae, 1.2 mm in length). The white waxy filaments are typical of the species. Photo courtesy of CABI, 2004.

Figure 2. Adult *A. spiniferus*. Photo courtesy of M.A. van den Berg. www.invasive.org

Adults: Adults are winged in both sexes, the females (1.7 mm long) being larger than the males (approximately 1.33 mm long). The wings are dark grey at ecdysis (Fig. 2), sometimes developing a metallic blue-grey sheen later; lighter markings on the wings appear to form a band across the insect. The body is orange to red initially; the thorax darkens to dark grey in a few hours. The limbs are whitish with pale yellow markings.

Pest Importance

Feeding by *A. spiniferus* causes general weakening and eventual death of heavily infested plants owing to sap loss and development of sooty mold (Anonymous, 1974). The sooty mold growing on the honeydew deposits block light and air from the leaves, reducing photosynthesis. USDA (1975) described it as "one of the most destructive Aleyrodids attacking citrus in tropical Asia and the seventh most important citrus pest in Japan." Clausen (1978) stated that *A. spiniferus* is a “common and at times serious pest of citrus and other plants in the Indo-Malaya region.” Xie (1993) described *A. spiniferus* as a pest of tea in China (Guangdong province). It was regarded as a serious pest of citrus in Micronesia until biological control agents were established (Suta and Esguerra, 1993). In India, *A. spiniferus* can be a serious pest of roses (David and Subramaniam, 1976).

Symptoms

Sticky honeydew deposits accumulate on leaves and stems and usually develop black sooty mold fungus, giving the foliage (even the whole plant) a sooty appearance (Fig. 3). Ants may be attracted by the honeydew. Infested leaves may be distorted. The insects are most noticeable as groups of very small, black spiny lumps on leaf undersides.

Figure 3. Sooty mold on citrus leaves. Photo courtesy of M.A. van den Berg. www.invasive.org

Known Hosts

Primary hosts

Citrus spp., *Rosa spp.* (roses)

Secondary hosts

*Psidium guajava (*guava), *Annona*, *Camellia sinensis* (tea), *Cinnamomum camphora* (camphor laurel), *Diospyros kaki* (oriental persimmon), *Eriobotrya japonica* (loquat), *Ficus racemosa* (cluster tree), *Hibiscus* (rosemallows), *Plumeria rubra var. acutifolia* (Mexican frangipani), *Pyrus pyrifolia* (Japanese pear), *Salix* (willow), *Sapium sebiferum* (Chinese tallow tree), and *Vitis vinifera* (grapevine)

Aleurocanthus spiniferus has not been recorded from as wide a host range as *A. woglum*i, but it feeds on members of 13 plant families and could potentially attack hosts in addition to those listed.

Known Distribution

Aleurocanthus spiniferus originated from southeast Asia and has spread to the tropical and subtropical regions of the Old World, overlapping with the distribution of *A. woglumi* in some areas. The orange spiny whitefly has spread to Africa, Australia, the Caribbean and the Pacific Islands. In the Caribbean Islands, orange spiny whitefly was recorded as an occasional pest of citrus in Jamaica. For the Pacific Islands, it was first recorded in Guam in 1951 where it was observed not only on citrus, but also on rose, grape, peach, pear and guava. Orange spiny whitefly was first detected on rose foliage in Honolulu, Oahu in 1974. Subsequent surveys discovered it on navel orange, lime, tangerine and pear, but infestations were reportedly low (USDA, 1974). In Africa, the first reports of this pest were in Swaziland (1987) and South Africa (1988). Established populations of orange spiny whitefly do not occur in the continental US (Gyeltshen and Hodges, 2005).

Both *A. spiniferus* and *A. woglumi* occur on citrus in Kenya, but have different distributions: *A. spiniferus* in the Central Highlands and *A. woglumi* at lower altitudes by Lake Victoria and the coast (Cock MJW, International Institute of Biological Control, Ascot, UK, 1990).

Potential Distribution within the US

Currently present in Hawaii.

Survey

Aleurocanthus spiniferus is most often found on citrus and roses. Leaves and steams should be closely examined, especially shrubs or trees, for signs of sooty mold or sticky honeydew or ants running about. A heavy infestation gives trees an almost completely black appearance. Look for distorted leaves with immature stages of *A. spiniferus* on the undersides. The adults fly actively when disturbed. Good light conditions are essential for detection; in poor light, a powerful flashlight is helpful. A large hand lens may be necessary to help recognition of the dorsal spines on immature stages.

Key Diagnostics

Several similar species of *Aleurocanthus* also occur on citrus, including *A. citriperdus* and *A. woglumi*. These species differ from each other in microscopic characters of the 'pupa' and

require expert preparation and identification to distinguish them. The main field characteristic difference between orange spiny whitefly and citrus blackfly, *A. woglumi*, is that the white wax fringe that surrounds their pupal case margins is generally twice as large for the orange spiny whitefly (Gyeltshen and Hodges, 2005).

Aleurocanthus woglumi

Scientific Name
Aleurocanthus woglumi Ashby

Synonyms:
Aleurocanthus punjabensis, A. woglumi var. formosana, Aleurodes woglumi

Common Name(s)
Citrus blackfly, blue grey fly, citrus spring whitefly

Type of Pest
Whitefly

Taxonomic Position
Class: Insecta, **Order**: Hemiptera, **Family:** Aleyrodidae

Reason for inclusion in manual
Western Region pest list

Pest Description
The following details are based on Smith et al. (1992). The elongate to oval eggs (0.2 mm long) are yellow when first laid and then darken to charcoal grey or black; each is attached to the leaf by a short pedicel. Eggs are laid in a spiral pattern on the underside of the leaf (Fig. 1A).

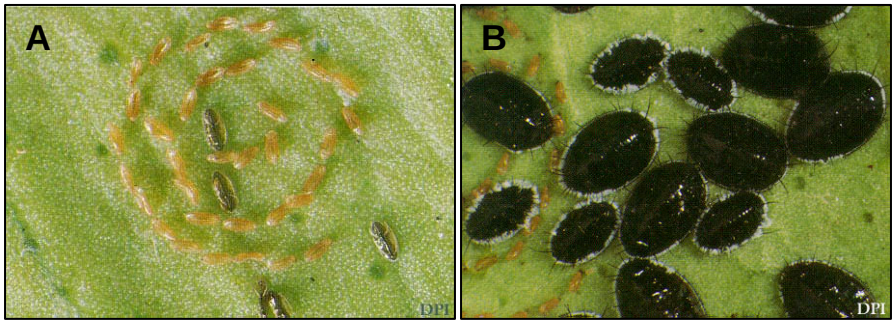


Figure 1. *A. woglumi* egg spiral and first instars (A) and pupae (B). Photos courtesy of the Florida Division of Plant Industry.

The six-legged, dusky, elongate first-instar larvae (0.3 x 0.15 mm) have two long and several shorter, slender dorsal glandular spines and are brown in color (Fig. 1A). All subsequent immature stages are sessile, have non-functional leg stubs and possess numerous, dark dorsal spines on which a stack of exuviae of earlier instars may occur. The second instar (0.4 x 0.2 mm) is a dark brown to charcoal convex disc with yellow markings, while the third instar (0.87 x 0.74 mm) is usually black with a rounded, greenish spot on the anterior part of the abdomen and obvious dorsal spines. In the fourth immature stage or 'pupa' (Fig. 1B), females are larger (1.25 mm long) than males (1 mm long). This stage is black, has numerous dorsal spines and is often surrounded by a white fringe of waxy secretion. This is the stage required for identification purposes.

Figure 2. Adult *A. woglumi* (larvae in background) showing metallic grey wings with light markings, red abdomen and white-yellow legs and antennae. Photo courtesy of CABI, 2004.

Adults are winged in both sexes, the females (1.7 mm long) being larger than the males (approximately 1.33 mm long). The wings are dark grey at ecdysis, sometimes developing a metallic blue-grey sheen later; lighter markings on the wings appear to form a band across the insect. The body is orange to red initially; the thorax darkens to dark grey in a few hours. The limbs are whitish with pale yellow markings (Fig. 2).

Pest Importance

Citrus blackfly infests over 300 host plants, and is suitable for large population development (Fig. 3). It damages citrus by sucking nutrients from foliage, which weakens the plants, damages new leaf growth, and reduces nitrogen levels in infested leaves. Sooty mold growing on honeydew blocks light and air from the leaves, reducing photosynthesis. This can reduce fruit set by up to 80% or more (Eberling, 1954). Crop losses of lime due to *A. woglumi* were recorded at 25% by Watts and Alam (1973). In Mexico, citrus blackfly is regarded as a threat to citrus crops and to other crops, such as mango, pear and coffee, which are grown adjacent to heavily infested citrus groves. *Aleurocanthuswoglumi* is a constant menace to citrus and other crops in the US and Venezuela. It has been recorded as seriously affecting citrus in India (David and Subramaniam, 1976). Le Pelley (1968) depicts it as a severe pest of coffee in the New World.

Figure 3. Heavy infestation of citrus blackfly on citrus leaves. Photo courtesy of Florida Division of Plant Industry.

Symptoms

Sticky honeydew deposits accumulate on leaves and stems and usually develop black sooty mold fungus, giving the foliage (even the whole plant) a sooty appearance. Ants may be attracted by the honeydew. Infested leaves may be distorted. The insects are most noticeable as groups of very small, black spiny lumps on leaf undersides (Fig. 3).

Known Hosts

Aleurocanthus woglumi is a polyphagous species with a strong preference for citrus. Unlike *A. spiniferus,* it is seldom found on roses. In Mexico, *A. woglum*i has been recorded on 75 species of host plant from 38 families (Shaw, 1950). Steinberg and Dowell (1980) found evidence suggesting that *A. woglumi* is incapable of infesting host species other than citrus for more than three generations, which may explain why serious infestations of other hosts are usually found in close proximity to citrus groves. *Aleurocanthus woglumi* has also been reported from *Croton spp.*

Primary hosts

Citrus spp.

Secondary hosts

Anacardium occidentale (cashew nut), *Annona*, *Ardisia swartzi*, *Averrhoa carambola* (carambola), *Buxus sempervirens* (box), *Carica papaya* (papaw), *Cestrum*, *Cocos nucifera* (coconut), *Coffea* (coffee), *Coffea arabica* (arabica coffee), *Cydonia oblonga* (quince), *Eugenia*, *Hibiscus* (rosemallows), *Laurus nobilis* (bay laurel), *Litchi chinensis* (leechee), *Mangifera indica* (mango), *Manilkara zapota* (sapodilla), *Morus* (mulberrytree), *Murraya*, *Musa* (banana), *Passiflora edulis* (passionfruit), *Persea americana* (avocado), *Plumeria* (frangipani), *Populus* (poplars), *Psidium guajava* (common guava), *Punica granatum* (pomegranate), *Pyrus* (pears), *Rosa* (roses), *Vitis*, and *Zingiber officinale* (ginger).

Known Distribution

Aleurocanthus woglumi originated from southern Asia and is widely spread throughout the tropical and subtropical regions, overlapping with the distribution of *A. spiniferus* in some areas. Currently, it is present in Bangladesh, Bhutan, Cambodia, China, India, Indonesia, Iran, Laos, Malaysia, Maldives, Myanmar, Nepal, Oman, Pakistan, Phillipines, Singapore, Sri Lanka, Thailand, United Arab Emeraites, Vietnam, Yemen, Kenya, South Africa, Swaziland, Tanzania, Uganda, Zimbabwe, Bermuda, United States (Florida, Texas, Hawaii), Antigua and Barbuda, Bahamas, and multiple Central American and South American countries.

Potential Distribution in US

Aleurocanthus woglumi was discovered in the western hemisphere in Jamaica (1913). It spread to Cuba in 1916, Mexico in 1935, and was detected in Key West, Florida in 1934. It was eradicated from Key West in 1937. It was rediscovered in Ft. Lauderdale, Florida in 1976. Citrus blackfly was detected in Palm Beach and Dade counties in 1977; Lee, Highlands and Brevard Counties in 1979; Manatee county in 1986; Polk County in 1989; Marion and Volusia Counties in 1991; and Alachua County in 1992. At present, it is widely spread over central and south Florida from Cross Creek to Key West. It is also present in Texas and Hawaii.

Survey

Examine plants, especially shrubs or trees, closely for signs of sooty mold or sticky honeydew on leaves and stems, and/or ants. Look for distorted leaves with immature stages on the undersides. Good light conditions are essential; in poor light, a powerful flashlight is helpful. A large hand lens may be necessary to help recognition of the dorsal spines on immature stages.

Key Diagnostics

Several similar species of *Aleurocanth*us occur on citrus, including *A. citriperdus*, *A. husaini* and *A. spiniferus*. These species differ from each other only in microscopic characters of the 'pupa' and require expert preparation and identification to distinguish them reliably.

Diseases

Bacterial/Mollicute Diseases

Candidatus Liberibacter africanus, *Ca.* L. asiaticus

Scientific Name
Candidatus Liberibacter africanus Monique Garnier, *Candidatus* Liberibacter asiaticus Monique Garnier

Common Name(s)
Citrus greening, huanglongbing, yellow dragon disease, yellow branch disease, likubin (immediate withering disease), mottle leaf disease, citrus dieback, citrus phloem degeneration

Type of Pest
Plant pathogenic bacterium

Taxonomic Position
Phylum: Proteobacteria

Reason for inclusion in manual
National pest list, National select agent list, and Regulated plant pest list

Pest Description
Citrus greening is caused by the fastidious, phloem-limited Gram-negative bacteria, *Candidatus* Liberibacter africanus in Africa, *Candidatus* Liberibacter asiaticus in Asia, *Candidatus* Liberibacter asiaticus and *Candidatus* Liberibacter americanus in Brazil. Despite many attempts, the citrus greening bacteria have not been grown in culture; thus, the bacterium is often referred to as *Candidatus* Liberibacter spp. The Asian form of the bacterium is considered heat-tolerant and the African form is considered heat-sensitive. Symptoms are not displayed when temperatures are above 25 to 30 °C for the African form, whereas the Asian pathogen displays symptoms at temperatures above 30 °C. Symptoms are also more severe with the Asian form.

Figure 1: Yellow shoots typical of citrus greening. Photo Courtesy of T.R. Gottwald and S.M. Garnsey

The disease is spread by vegetative propagation (grafting), experimentally by dodder (*Cuscuta spp*.), and by two phloem-feeding psyllid vectors. The Asian citrus psyllid, *Diaphorina citri* Kuwayama, and the African citrus psyllid, *Trioza erytreae* (del Guercio), vector the Asian and African species, respectively. Either of the two psyllid vectors has proven to be capable of hosting either pathogen in the laboratory; however, it is not known whether this occurs in nature (da Gracca, 1991). The bacterium can be seen by electron microscopy in the sieve tubes of infected trees and in vectors as elongated sinuous rods, 0.15 to 0.25 μm in diameter. The bacterium's resistance to culture on artificial media has made the study of the organism's population dynamics, epidemiology, and interactions with its psyllid vectors quite difficult. The disease may be seed transmitted.

In 2004, citrus greening disease was reported in the main citrus growing areas of São Paulo, Brazil. The bacteria from citrus leaves had 93.7% similarity with the Asian and African form of the citrus greening pathogen. In 2005, it was reported that the bacterium found in São Paulo was sufficiently different from known Liberibacters to warrant a new species named *Candidatus* Liberibacter americanus (Colletta-Filho et al., 2004; Texeira et al., 2005.) The vector, *Diaphorina citri,* is established in Brazil.

Pest Importance

Citrus greening is thought to have originated in the Indian subcontinent. Globally, greening has been regarded as one of the most important threats to commercial and sustainable citrus production. Losses due to greening are not easy to assess. Sometimes only sectors of a tree are affected and losses are small; in other cases, the entire tree is infected and crop loss is total (de Graca, 1991). In some areas of the world where the disease is endemic, citrus trees decline within five years of planting, and most will not bear usable fruit. Such losses are significant, since profits are only attainable 8 to 10 years after planting. In countries where greening occurs, it is the primary limiting factor for citrus production. Citrus greening has a long incubation period and many latently infected citrus plants occur in the field (McClean, 1970). Plants may, therefore, be infected with citrus greening and not show symptoms. If latently infected plants are used in propagating and grafting, the disease is spread.

Figure 2: Close up of a yellow shoot
Photo Courtesy of R.F. Lee

Control measures are limited to the use of disease-free propagating stock, rouging of infected trees, and chemical or biological control of the vectors. The vector *D. citri* was found in Florida in 1998, in Texas in 2001, and has spread considerably since its introduction (Knapp et al., 2004; French, 2002). The presence of the vector in the United States, and the recently reported outbreaks of citrus greening in Brazil, raises concerns that any

Figure 3: Blotchy, mottled leaf
Photo Courtesy of R.F. Lee

unrecognized introduction of greening-infected citrus, in the past or future, could lead to the introduction of the pathogen. Citrus greening disease was recently (September 2005) detected in Miami-Dade County; few details are available at this time.

Symptoms

Citrus greening is a systemic disease. The first visible symptom of greening is often a diagnostic blotchy mottle on new growth somewhere in the canopy (Fig. 1,2,3). Characteristics of greening can occur in one branch or part of the tree to first shows symptoms; it then progressively spreads throughout the tree canopy. As the disease progresses, trees turn chlorotic, develop twig dieback, and rapidly decline to a nonproductive state.

Figure 4. Zinc deficiency symptoms on citrus. Photo courtesy of S. Futch

Affected leaves are blotchily mottled (Fig. 3), pale yellow, or have the appearance of foliage affected by zinc and other nutrient deficiencies. Leaves with citrus greening have a mottled appearance that differs from nutrition-related mottling in that greening-induced mottling usually crosses leaf veins. Nutrition related mottles are usually found between or along leaf veins. Leaves on citrus greening-affected trees may be small and upright (Fig. 4). Leaves with citrus greening may have prominent yellow veins (Fig. 5). The leaves may prematurely fall off.

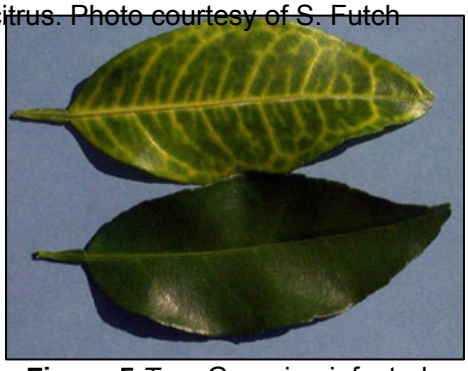


Figure 5 *Top:* Greening infected leaf with yellow vein symptom. *Bottom:* Healthy citrus leaf. Photo Courtesy of R.F. Lee

The fruit are usually small, misshapen, and sour to bitter in taste, which is in contrast to fruit affected by severe citrus tristeza virus (CTV), blight, or stubborn, where the fruits are sweet. The seeds within the fruit are often aborted. The fruit fails to color uniformly as it ripens (stylar end greening) (Fig. 6). On Mandarin orange, the fruit develops with uneven ripening in that it appears half orange and half green; this symptom is responsible for the common name 'greening' and primarily occurs with the African species and cool climates. In Brazil, affected fruit develop a blotchy mottle.

Figure 6: Stylar end greening on mandarin orange fruit. Photo Courtesy of T.R. Gottwald and S.M. Garnsey

Known Hosts

Citrus greening is a disease of rutaceous plants. The citrus greening bacterium generally infects citrus (EPPO, 1988). The bacterium may persist and multiply in most *Citrus* spp.; most severe symptoms are found on oranges (*C. sinensis*),

mandarins (*C. reticulata*), and tangelos (*C. reticulata* x *C. paradisi*). Somewhat less severe symptoms are found on lemons (*C. limon*), grapefruits (*C. paradisi*), *C. limonia*, *C. limmettioides*, rough lemons (*C. jambhiri*), kumquats (*Fortunella spp.),* and citrons (*C. medica*). Symptoms are even weaker on limes (*C. aurantiifolia*) and pummelos (*C. grandis*). A range of other rutaceous plants, including ornamentals and wild species, are susceptible to infection; however, the severity of symptoms displayed varies. Hosts include *Poncirus, Severinia buxifolia* (box thorn) *Lemonia acidissima, Murraya*, *Toddalia*, and *Calodendrum capense*. The citrus greening bacterium has been experimentally transmitted by *Cuscuta campestris* (dodder) from citrus to tow non-rutaceous hosts, *Catharanthus roseus*, and tobacco (Garnier and Bove, 1983).

Known Distribution

Greening caused by *Ca.* L. asiaticus is found in many countries throughout Asia and is spreading in Indonesia. *Candidatus* Liberibacter africanus occurs in numerous countries in Africa and is also spreading. Both *Ca.* L. asiaticus and *Ca.* L. africanus were present in Reunion and Mauritius, sometimes co-existing in the same tree (Garnier et al., 1996). The greening bacterium and its vectors have not been detected in Australia. *Candidatus* Liberibacter americanus was detected in Brazil in 2004.

Asia: Bangladesh, Bhutan, Cambodia, China, India, Indonesia, Japan, Laos, Malaysia, Myanmar, Nepal, Pakistan, Papua New Guinea, Phillipines, Saudi Arabia, Taiwan, Thailand, Vietnam, and Yemen (EPPO, 1988).

Africa: Burundi, Cameroon, Central African Republic, Comoros, Ethiopia, Kenya, Madagascar, Malawi, Mauritius, Rwanda, Reunion, Somalia, South Africa, Swaziland, Tanzania, and Zimbabwe.

South America: Brazil

Potential Distribution within the US

Areas within the United States with citrus production and temperatures above 25 °C would be at risk for greening disease. The African species of Liberibacter only occurs in cool climatic conditions (below 25 to 30 °C or 77 to 86 °F), at elevations above 600 to 1000 m, where the primary vector, *T. erytreae,* is most numerous (Green and Catling, 1971). In contrast, the Asian species occurs at low elevations with a hot climate (above 30 °C or above 86 °F). The psyllid vector is *Diaphorina citri*. Among the environmental factors, strong wind seems most important (Koizumi et al., 1997). *D. citri* can fly, but only within a short range, such as from leaf to leaf or twig to twig, when observed in an incubator. A strong wind may disseminate the infected psyllids from the donor to the receptor in a leeward direction, which enhances disease spread (Koizumi et al., 1997).

In September 2005, the greening bacterium was detected in Miami-Dade County, Florida. Few details concerning the extent of the infection are available at this time.

Survey Procedure

Currently, there are no specific survey methods for this disease. Greening is commonly identified in the field by foliage and fruit symptoms. A yellowing of the tree canopy, blotchy mottled leaves and small lopsided fruits with aborted seeds provide the best indication of

citrus greening infection. Further diagnosis requires indexing on susceptible citrus seedlings by graft inoculation or confirmation through the use of DNA hybridization or polymerase chain reaction (PCR) to identify bacterium.

Symptoms of greening disease also may be confused with stubborn disease (*Spiroplasma citri),* citrus tristeza closterovirus (CTV) infection, *Phytophthora* infection, citrus blight, or certain nutrient deficiencies; as a result, greening disease difficult to identify in the field. The blotchy mottle, combined with the bitter, lopsided fruit, is helpful to differentiate greening from citrus blight, CTV, and citrus stubborn.

The first visible symptom of greening is usually a branch with small leaves that have a blotchy mottle, followed by a single yellow shoot in the canopy, which then progressively spreads throughout the tree canopy. Initial survey efforts should focus on the tree canopy. If symptoms occur in the canopy, then the leaves and fruits should be examined for further symptoms.

Alternative hosts always play a role in an epidemic disease, but they are often neglected in epidemiological studies, especially when they cannot be easily recognized. The greening bacterium can replicate in boxthorn (*Severinia buxifolia*) and wood apple (*Limonia acidissima*), but not in orange jasmine (*Murraya paniculata* var. *paniculata*) and curry leaf (*Murraya euchrestifoli*a) (Hung et al., 2000). Orange jasmine, and curry plant to a lesser extent, are common hosts for the psyllid vectors, particularly *D. citri*, but not for the greening organism. These hosts also should be monitored for the bacterium and the vector during surveys, particularly if located in close proximity to a citrus producing area.

Key Diagnostics

The bacterium that causes citrus greening is difficult to detect because of its low concentration and uneven distribution in its natural hosts (da Graca, 1991). Fortunately, the application of DNA probes shows promise for overcoming the difficulty of greening detection.

Spiroplasma citri

Scientific Name
Spiroplasma citri

Common Names
Stubborn, citrus stubborn disease, little leaf (Israel), acorn disease.

Type of Pest
Plant pathogenic Mollicute

Taxonomy
Class: Mollicutes, **Family:** Spiroplasmataceae

Reason for inclusion in manual
Disease that could be confused with citrus greening

Pest Description
Saglio et al. (1973) characterized the organism and proposed the name of *Spiroplasma citri*. It is chemo-organotrophic, facultative anaerobic, and has wall-less pleomorphic cells with a characteristic spiral morphology (Fig. 1). The minimum viable length of a helix is two turns (2.0µm x 0.1 to 0.2 µm). The helices are motile by flexing or rotation, apparently mediated by a contractile fibrillar cytoskeleton bound to the inner surface of the spiroplasmal membrane (Fig. 1). Some strains are non-motile and non-helical. *Spiroplasma citri* is one of the very few plant pathogenic mollicutes that can be cultured (Saglio et al., 1973; Bradbury, 1991).

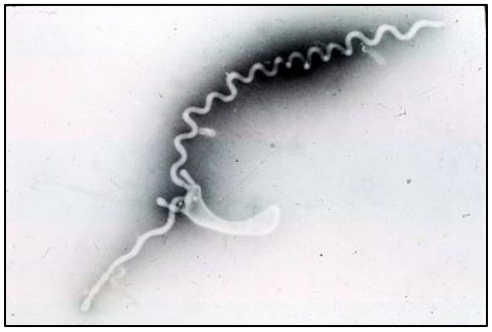


Figure 1. *Spiroplasma citri*. Note the spiral-helical nature of the organism as seen in the dark-field microscope. Photo Courtesy of EcoPort (http://www.ecoport.org)

Spiroplasma citri infects the phloem sieve tubes of its hosts. The pathogen persists in affected trees as they decline. It is an obligate parasite, surviving in citrus or in a variety of other host plants. It is naturally transmitted by phloem sap-feeding leafhoppers: *Circulifer tenellus*, *Scaphytopius nitridus* and *S. acutus delongi* in California (Oldfield, 1988); and by *Neoaliturus haematoceps* and *C. tenellus* in the Mediterranean area (Bove, 1986; Klein et al., 1988). None of these vectors have a particular preference for citrus as a host, and it is likely that they acquire *S. citri* from other hosts. *Spiroplasma citri* multiplies in its insect vectors, which become infective after 10 to 20 days of acquisition feeding (Liu et al., 1983). The insects are able to infect their hosts throughout their lives (which may be shortened by the infection); there is no transovarial transmission.

Spiroplasma citri develops best in citrus under hot conditions (28 to 32°C). It has not been a problem in cool areas or in areas with warm and humid climates. *Spiroplasma citri* may not produce any conspicuous symptoms at lower temperatures. *Spiroplasma citri* from citrus-free areas can be experimentally vector-transmitted to citrus (Gumpf, 1988); the pathogen from citrus can be experimentally transmitted to horseradish (Sullivan et al., 1987). There is no indication of special races or strains attacking citrus.

Pest Importance

Spiroplasma citri causes a citrus stubborn disease of citrus and can greatly reduce the quality and quantity of the yield under hot, dry conditions. In California, the main economic hosts are orange, grapefruit and tangelo, of which 5 to 10% of trees are estimated to be affected. Recent research indicates that citrus stubborn is probably more widespread in California than previously thought; as a result, economic losses are also greater (Rangel et al., 2004). In the Mediterranean area, especially in Syria, stubborn is very serious due to the abundance of the vector *Neoaliturus haematoceps*, and the rapid re-infection of new, healthy budwood (Bove, 1986).

Figure 2. Symptoms of stubborn in California. Note the characteristically stunted and compressed appearance of a stubborn-infected navel orange tree (right) in comparison with a non-infected tree of the same age (left). Photo Courtesy of EcoPort (http://www.ecoport.org)

Although *S. citri* naturally infects many hosts, some of which are crop plants, it does not have any economic impact on them. The main significance of these hosts would be as reservoirs inoculum of *S. citri*.

Symptoms

Stubborn is rarely lethal in citrus, but young trees affected are often severely stunted (Fig. 2) and give low yields. Leaves are shorter and broader, cupped, abnormally upright, sometimes mottled or chlorotic (Fig. 3). Under very hot conditions, leaves on some shoots may have misshapen, blunted or heart-shaped yellow tips. Shoots may be abnormally bunched and the development of multiple axillary buds may give rise to witches' brooms or ‘crazy top’ (Fig. 3.). Infected trees may flower out of season.

Figure 3. Symptoms of stubborn (left) and the “crazy top” symptom on several branches. Photo Courtesy of R. F. Lee

Fruiting is typically suppressed by infection. Fruits are infected at all stages of development and may be stunted, lopsided or acorn-shaped, due to a thick rind at the base and thin rind at the tip. In severe cases, the albedos of sweet orange and grapefruit have a blue color. Fruit may fail to green (e.g., the stylar end remains green while the peduncular end discolors) (Fig. 4). Seeds may be partially aborted. The name 'stubborn' arises from the persistence of the original characters of an infected citrus tree when it is 'top-worked' with healthy budwood (Bove and Garnier, 2000; CABI, 2005).

Figure 4. Symptoms of stubborn on Navel oranges in California. Note small, asymmetric, and unevenly colored fruits (stylar-end greening) as compared to the two healthy fruit on the bottom. Photo Courtesy of EcoPort (http://www.ecoport.org)

Known Hosts

The principal economic hosts of *S. citri* are citrus species. Orange, lemon, mandarin and grapefruit are particularly susceptible. Acid limes, trifoliate orange, and trifoliate orange hybrids can be experimentally infected by graft inoculation (Garnsey and Gumpf, 1988). The greatest number of host plants is in the Brassicaceae. All susceptible plants, except those in the families Rosaceae and Rutaceae, are herbaceous.

Numerous cultivated and wild plants have been found to be naturally infected by *S. citri* without showing symptoms; the incidence of the disease in these plants seems to be too low to have attracted attention. The only recognized disease caused by *S. citri* on a non-rutaceous host is brittle root of horseradish (Armoracia lapathifolia) in the eastern US (Sullivan et al.,1987). London rocket (*Sisymbrium irio*) and periwinkle (*Catharanthus roseus*) are commonly used as experimental hosts for phytoplasmas and are naturally infected in Mediterranean countries.

Known Distribution

Stubborn disease of citrus was first described in 1915 in Washington Navel trees near Redlands, California (Rangel et al., 2004). It was later found in 1928 in Palestine. This disease has not been reported in eastern Asia, the center of origin for citrus, or tropical Africa. It is not clear whether *S. citri* is indigenous or introduced in North America. Its main vectors are located in the southwestern US, and include *Circulifer tenellus* and *Scaphytopius nitridus*. The leafhopper, *C. tenellus* (primarily a sugarbeet insect), is of Mediterranean origin (CABI, 2004).

Stubborn has been found in Algeria, Cyprus, Egypt, France (Corsica), Greece, Iran, Iraq, Israel, Italy (Sardinia, Sicily), Jordan, Lebanon, Libya, Morocco, Pakistan, Saudi Arabia, Spain, Syria, Tunisia, Turkey, and Yemen. It is also present in Mexico. Records from South America (Argentina, Brazil, Peru, Suriname, Venezuela) are based on symptoms only and should be considered as unconfirmed (CABI, 2004).

Potential Distribution in the US

Stubborn is well-established in most warm, dry, inland citrus producing areas in Arizona and California (Rangel et al., 2004). For unknown reasons, the disease has not been found in

Florida or Texas (Olsen et. al., 2000). The hot, dry weather in the San Joaquin and Desert Valley regions has favored the development and spread of the stubborn pathogen. Stubborn primarily affects sweet orange, grapefruit, and tangelo trees. In Arizona, stubborn is especially prevalent in Washington Navel trees (Olsen, et al., 2000). The disease is more of a problem in young orchards than in mature groves. New spread is likely to occur via infected budwood. Infective vectors may be carried on citrus plants, but the insects concerned are mobile and do not preferentially feed on citrus; thus, the risk is considered minimal.

Survey

Currently, there is no specific survey method published for this disease.

The classic diagnostic approach is graft inoculation of highly sensitive plants, such as orange cv. Madame Vinous, kept at 32°C in the day and 27°C at night. Other indicators used are grapefruit cv. Marsh and tangelo cv. Sexton. Inoculation is usually by side graft, which is more difficult than regular inoculations (Rangel et al., 2004). Symptoms develop in 2 to 3 months. The best inoculum is obtained from young leaf patches including the midrib.

A definitive diagnostic method is to isolate and culture *Spiroplasma citri* on artificial medium. The pathogen forms the typical umbonate or “fried egg” colonies on solid medium (Lee and Davis, 1984). Its helicity can be confirmed by dark field microscopy and its identity by serological methods or PCR. *Spiroplasma citri* can be isolated from shriveled and discolored seeds, the peduncular end of the fruit axis, or mottled summer leaves (Bove et al., 1084). Since the organism can be cultured, antisera is relatively easy to obtain (spiralin is the most abundant membrane protein and the major surface antigen), and ELISA can be used for the detection and identification of *S. citri* in extracts from plants and insects (Bove et al., 1984). Ranger et al. (2004) developed a PCR-based technique to rapidly detect *S. citri*. Prior to PCR amplification, collected tissue is surface-sterilized and ‘inoculated’ into liquid media. DNA is extracted from a sub-sample of 0.5 ml of the original 3 to 5 ml of media, and used for the PCR reaction utilizing the primers published by Foisaac et al. (1996). Detection of *S. citri* is possible from bark (budwood) and fruit, preferably collected during the warmer months.

Xanthomonas axonopodis pv. citri

Scientific Name
Xanthomonas axonopodis pv. citri Hasse

Synonyms:
Xanthomonas campestris pv. citri, *X. citri*, X*. citri f.sp. aurantifolia*

Common Name(s)
Citrus canker, citrus bacterial canker

Type of Pest
Plant pathogenic bacterium

Taxonomic Position
Phylum: Proteobacteria, **Order:** Xanthamonadales, **Family:** Xanthamonadaceae

Reason for inclusion in manual
National pest list, Eastern Region pest list, Emerging plant pest list, and Regulated plant pest list

Pest Description
Xanthomonas axonopodis pv. *citri* is a straight, rod-shaped, obligate aerobic gram-negative bacterium, measuring 1.5 to 2.0 x 0.5 to 0.75 µm. It is motile by means of a single polar flagellum. Colonies on agar plates (NA or PDA) with glucose are creamy-yellow with copious slime. The yellow pigment is a xanthomonadin.

Different strains of citrus canker have been recorded throughout the world. The Asiatic form, or A-strain, is the most widespread and severe. There are also different forms of A-strain distinguished by host range, cultural and physiological characteristic, and molecular biology techniques.

Figure 1. Lesions of citrus canker on leaves. Photo courtesy of T. Riley

Pest Importance
Citrus canker is by far the most serious disease of commercial citrus varieties and citrus relatives. Its presence in a particular area can cause devastating socioeconomic and political impacts. Yield losses due to citrus canker may result from defoliation, premature fruit drop and blemished fruits, but losses are primarily due to a loss of markets due to quarantines on the transport, sale, and export from affected areas.

Symptoms

All young above-ground tissues are susceptible to the pathogen. The pathogen enters plant tissues through stomates or wounds, particularly those caused by the citrus leafminer (*Phyllocnistis citrella*). On leaves, lesions develop as light yellow, raised, tiny blister-like lesions on the lower surface (Fig. 1). As the lesions enlarge, they become corky, crateriform, turn tan to brown with a water-soaked margin surrounded by a chlorotic halo. Canker lesions are roughly circular, and vary in size from 5 to 10 mm, depending on the susceptibility of the host plant. The center of lesions dry out and may fall out producing a shot hole effect.

Symptoms of the disease on twigs and fruits are similar to those in leaves (Fig. 2, 3). Lesions on twigs on angular young shoots provide the perpetuating inoculum. *Xanthomonas axonopodis* pv. *citri* is disseminated by rainwater running over the surfaces of lesions and splashing onto uninfected shoots. Rainstorms, such as typhoons and hurricanes, encourage outbreaks of citrus canker. Although rainstorms can transport bacteria up to 100 m or more in small raindrops and/or aerosols, effective infection rarely occurs more than a few rows downwind.

Figure 2. Lesions of citrus canker on stem. Photo courtesy of T. Riley.

Known Hosts

The pathogen affects all cultivars of the genus *Citrus* and some members of the family Rutaceae, such as *Poncirus*, *Fortunella* and *Citrofortunella*. Among commercial citrus varietes and rootstocks, citrus canker is most severe on grapefruit (*C*. x *paradise*), Key limes (*C*. *aurantiifolia*), Palestine sweet lime (*C*. *limettioides*) and trifoliate orange (*Poncirus trifoliate*) and their hybrids. Some types of mandarins (*C*. *reticulate*) are considered resistant, whereas calamondins (X *Citrofortunella microcarpa*) and kumquats (*Fortunella* spp.) are reported to be highly resistant.

Figure 3. Lesions of citrus canker on fruit. Photo courtesy of T. Riley

Known Distribution

The Asian or A-strain of *Xanthomonas axonopodis pv. citri* is thought to have originated in southeastern Asia or India, and spread to Japan, New Zealand, Australia, southern and central Africa, Middle East, South America, and southeastern the United States (Florida).

Potential Distribution within the US

Citrus canker is not present in all citrus-growing regions where the climate is conducive for its development. Currently, it is present in Florida. In Florida, citrus canker is subject to intensive surveys; eradication programs are in progress in both residential and commercial groves.

Survey

Since *X*. *axonopodis* pv. *citri* can infect any green part of the plant, including leaves, twigs and fruits of host plants, these plant parts should be closely examined during any survey. Citrus canker occurs in any season on seedlings and young trees in which there is a flush of abundant angular shoots. As a general rule, the pathogen is capable of infecting green citrus tissues while they are in the expansion phase of growth. The disease becomes sporadic once leaves, twigs, and fruits reach full development. In Florida, almost all infections on leaves and stems occur during the last half of the expansion phase.

The USDA researchers at the U.S. Horticultural Research Laboratory in Fort Pierce, FL., have developed the sentinel tree survey method. In this method, each square mile is divided into 12-by-12 grid of 144 subsections. A sentinel tree is selected and tagged in each subsection and used as an early warning system for new canker outbreaks. Sentinel trees are visually examined every 30 to 90 days.

In commercial fields, surveys may include all trees or any combination of rows, for example, trees in high hills in every other two rows. Trees can be examined only in the high hill side or both in the high hill and low hill side.

Key Diagnostics

Leaves have raised rough lesions surrounded by brown water-soaked margins with a bright yellow halo. Sun shining through the leaves highlights the yellow halos. Citrus leprosis virual lesions are usually very characteristic, but may sometimes be mistaken for lesions of citrus canker caused by the bacterium *Xanthomonas axonopodis* pv. *citri.* An abnormal leaf fall is also indicative of the disease. Fruits are often blemished and drop prematurely. Lesions on stems are similar to those on leaves (1 mm deep), and twigs often dieback.

Although citrus canker can be diagnosed in the field based on its unique symptoms, identification of the causal agent must be confirmed by inoculation tests to susceptible host plants or by molecular methods.

Xylella fastidiosa

Scientific Name
Xyllella fastidiosa Wells

Common Name(s)
Citrus variegated chlorois, amarelinho (Brazil), pecosita (Argentina)

Type of Pest
Plant pathogenic bacterium

Taxonomic Position
Phylym: Proteobacteria, **Order:** Xanthamonadales, **Family:** Xanthamonadaceae

Reason for inclusion in manual
National pest list, National select agent list, and Eastern Region pest list

Pest Description
Xylella fastidiosa is a fastidious, gram-negative, rod-shaped, xylem-limited bacterium with rippled cell walls. It is strictly aerobic, non-flagellate, does not form spores, and measures 0.4 x 4 μm (Hartung et al., 1994). The bacteria are tightly packed in the lumen of xylem vessels when viewed by electron microscopy (Fig. 1) and are transmitted in a persistent manner by various leafhopper species. The vessels are ultimately blocked by bacterial aggregates and by tyloses and gums formed by the plant.

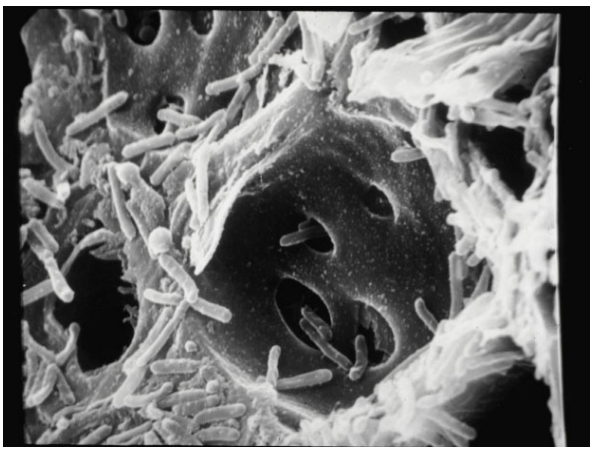


Figure 1. *X. fastidiosa* in xylem vessels. Photo courtesy of D.R. Cook

This taxon currently includes at least three different pathogen groups/strains. Bacteria in one group, the group of greatest concern, are known to infect citrus (citrus variegated chlorosis) and coffee (coffee leaf scorch) in South America, and were recently found in Costa Rica. Another strain infects plums in South America (plum leaf scald). The other groups occur in North America, where the diseases have been known for many years. The bacteria in the North American groups infect many plants, including grapes (Pierce's disease), alfalfa (alfalfa dwarf), peach (phony disease), almonds (almond leaf scorch), and, additionally, cause scorch diseases in a number of different shade trees (sycamore, oak, and maple). One distantly related strain causes pear leaf scorch in Taiwan (Horvath, 2005).

Pest Importance

Figure 2: Citrus variegated chlorosis . Photo courtesy of M.J.G. Beretta

Citrus variegated chlorosis (CVC) (Fig. 2), a new disease primarily of sweet orange (*Citrus sinensis*), has been associated with strains of *X. fastidiosa* in Brazil (Chang et al., 1993; Hartung et al., 1994). The disease was first observed in Argentina during the early 1980s and thereafter in Brazil (Roberto et al., 2002). The author's initial reports suggested that the disease posed an immediate threat to the Brazilian and world citrus industry. In 1996, the Brazilian state of Sao Paulo was responsible for 83% of the national citrus production; CVC was present in all citrus growing areas of Sao Paulo (Lopes et al., 2000). In the year 2000, 35% of the 200 million sweet orange trees in Sao Paulo showed CVC symptoms, representing a direct loss of more than US$100 million (Li et al., 2002). It has been estimated that in the absence of remedial measures, a CVC incidence of 90% in a grove could occur 12 years after introduction of a single infected tree; as a result, an individual tree may become unproductive within three years (Gottwald et al., 1993). The severity of the CVC disease problem contributed to the selection of a citrus strain of *X. fastidiosa* as the first plant pathogenic bacterium to have its entire genome sequenced (Simpson et al., 2000).

Epidemiological studies suggest that most CVC is spread from tree to tree within citrus groves. The bacterium is spread from plant to plant by grafting with infected bud material, by natural root grafts, and by sharpshooter leafhoppers (Hemiptera Cicadellidae). Because the bacterium is restricted to the xylem, transmission through budwood is low, but nevertheless, it is sufficient for the widespread distribution of the disease. In Brazil, 12 of 16 sharpshooter species tested were able to transmit the bacterium. In the U.S., the blue-winged sharpshooter (*Oncometopia nigricans*) has been experimentally shown to be a vector of the CVC strain of *X. fastidiosa*. The glassy winged sharpshooter (GWSS) (*Homalodisca coagulata*) has been observed transmiting Oleander Leaf Scorch (another *X. fastidiosa* strain) with more than 80% efficacy. The bacterium has recently been shown to be seedborne, which may require the monitoring and testing of seed and budwood sources and limiting the movement of fruit from infected locales (Li et al., 2003).

Pierce's disease, caused by another strain of *X. fastidiosa*, already limits grapevine production in the southeastern US and Central America. Citrus and coffee strains of *Xylella fastidiosa* from Brazil have been shown to induce Pierce's disease of grapevine (Li et al, 2002). Any future introduction of the CVC strains of *X. fastidiosa* into the United States pose a threat to both the sweet orange and the grapevine industries, given the high populations of sharpshooters found in many areas of Florida and California. The Pierce's disease strains of *X. fastidiosa* apparently are not capable of inciting CVC, as citrus has been commercially grown in California and Florida for more than a century in the presence of Pierce's disease infected grapes (Li et al., 2002).

Symptoms

Early symptoms of CVC include a foliar interveinal chlorosis (yellowing) resembling zinc deficiency on the upper surface of young of leaves as they mature (Fig. 3). As the leaves mature, small light-brown gummy lesions develop on the lower surface, corresponding with the chlorotic areas on the upper surface. Lesions become dark-brown or even necrotic, enlarging with time. The leaves are often smaller than normal. Very young leaves do not show symptoms. The English name of citrus disease, CVC, comes from the striking chlorotic variegation induced on sweet orange leaves by the pathogen (Fig. 4).

Figure 3: Leaf interveinal chlorosis. Photo courtesy of A. Purcell. www. invasive.org

Figure 4: Closeup of interveinal chlorosis. Photo courtesy of R. F. Lee

Initially, symptoms are present in some branches, but over time, the entire canopy displays the disease. Young, non-fruit bearing trees become systemically infected at a rapid pace, and generally display more severe symptoms than older trees. Trees 8 to 10 years old are not usually totally affected, but rather have symptoms on the extremities of branches. Infected trees show stunting and reduced growth rates, with twig and branch dieback and canopy thinning.

Blossom and fruit set occur at the normal time, but the usual fruit thinning does not occur, resulting in clusters of 4 to 10 early-maturing small fruit. Although sweet orange trees affected by the disease do not die, fruit from the trees may be severely undersized (Fig. 5), have hard rinds, lack juice, are acidic, and are of no commercial value. The rinds are hard enough to damage commercial juice extractors, causing entire lots of fruit to be rejected at processing plants (Lee et al., 1991). Sugar content of the fruit is higher than non-affected fruit, and typically ripens earlier than normal. Such fruit is unacceptable for either the juice or fresh market.

Figure 5: Healthy fruit (left) and CVC infected fruit (right). Photo courtesy of A. Purcell. www.invasive.org

One of the challenges in identifying this pathogen results from the relatively long latent period between infection and resulting symptoms. Typical symptoms do not develop from 9 to 12 months after infection in the field and symptoms can easily be mistaken for a zinc deficiency problem. Symptom expression

and incidence of CVC appear to be greater in warmer climates.

Known Hosts

The primary host of the CVC strain of *X. fastidiosa* is citrus. All sweet orange varieties are highly susceptible, with limes and grapefruit being less susceptible. The sweet orange cultivars, Pera, Hamlin, Natal, and Valencia, appear to be extremely susceptible to CVC. Lemons, mandarins, and some mandarin hybrids range from susceptible (show leaf symptoms) to tolerant (very mild or no leaf symptoms) to resistant (no leaf symptoms and no detectable bacteria). Coffee (*Coffea arabica* and *C. canephora* var. *robusta*) plants are susceptible; CVC bacteria cause the disease known as coffee leaf scorch. Several scientists have suggested that coffee may be the original host of this strain, with the bacteria being moved by vectors and adapting to citrus in nearby plantings.

Many strains of *X. fastidiosa* have wide natural host ranges; alternate hosts may serve as a major source of inoculum. The following (symptomless) weeds were collected in citrus groves in Brazil and found to be naturally-infected with the CVC strain of *X. fastidiosa* by polymerase chain reaction (PCR) testing: *Alternanthera tenella, Commelina benghalensis, Bidens pilosa, Euphorbia hirta, Brachiaria decumbens., Cenchrus echinatus, Digitaria horizontalis, Digitaria insularis, Spermacoce latifolia,* and *Solanum americanum* (Horvath, 2005). The following plants are hosts when mechanically inoculated: grape (*Vitis vinifera*), Madagascar periwinkle (*Cantharanthus roseus*), tobacco (*Nicotiana tabacum*), alfalfa (*Medicago sativa)*, and the weeds *Brachiara plantaginea* and *Echinochloa crus-galli* (Li et al, 2002; Lopes et al., 2000; Lopes et al., 2003; Monteiro et al., 2001).

Known Distribution

Argentina, Brazil, Costa Rica, Paraguay

Potential Distribution within the US

Citrus production areas with large populations of the sharpshooter vectors are at considerable risk, particularly Florida and California. In Florida, the sharpshooter *Oncometopia nigricans,* is native to the area, feeds on citrus, and is capable of transmitting *X. fastidiosa* (Brlansky et al., 2002). Currently, studies are underway to assess the potential of the glassy winged sharpshooter (GWSS), which is present in Florida and California, to transmit the CVC bacterium, which is a highly efficient vector of the oleander strain of *X. fastidiosa* (Brlansky et al., 2002). Recent epidemiological data showed that proximity to citrus increased the incidence and severity of Pierce's disease of grapevines in the Temecula Valley of California (Perring et al., 2001). This relationship occurs because in California the GWSS preferentially feeds and reproduces to high levels on citrus.

Survey (Taken from Horvath, 2005).

While CVC may occur on all cultivars, species, and hybrids of citrus, symptoms vary depending on the particular plant and its age. Sweet oranges are the most susceptible and show the most severe symptoms. Grapefruit, limes, mandarins, and mandarin hybrids show less severe symptoms. Rangpur lime, lemons, citron and pummelo are tolerant to the disease, showing very mild (if any) symptoms. Young infected trees generally show more severe foliar symptoms than mature infected trees.

Infected trees can randomly occur throughout the grove (orchard), making it necessary to visually inspect all trees for the presence of symptoms. Symptoms may occur on only one or a few branches, or (in later stages) throughout the canopy of the tree. The best symptoms are likely to be seen on the newest fully-expanded leaves. Please keep in mind that foliar symptoms can resemble zinc deficiency symptoms, especially in the early stages of the disease. If the grove has a history of zinc deficiency, it will be necessary to check for the gummy lesions on the undersides of the leaves when interveinal chlorosis is observed. When in doubt, always collect and submit samples for laboratory analysis. Typical fruit symptoms of the disease can be easily recognized from a distance.

Sampling Procedures: Collect at least 10 to 12 leaves of various ages showing typical symptoms, leaving the petioles attached. If available, include leaves that show the gummy brown lesions on the undersides. Cut 7 to 8 green twigs, 6 to 8 inches long from those areas of the suspect tree that show the best symptoms. Samples should be placed in plastic bags (place the leaves between dry paper towels and bundle the twigs together with a rubber band), along with their identifying numbers and the necessary collection and contact information. Double bag the samples. Mark the sampled trees and draw a map of the immediate area showing tree locations so that the trees can be found in the future, if necessary. Flagging the sampled branch or branches will make it easier to obtain additional samples if needed by the laboratory at a later date. Flagging or other markers attached to the trees help, but may become detached. Disinfect the pruning shears after sampling each tree with a bleach solution. Keep the samples as cool as possible, but do not freeze. Contact a diagnostic laboratory by phone prior to shipping the samples via overnight delivery service.

Commercial Grove Survey Procedures: It will be necessary to walk every row in the grove in order to inspect as much of the newer growth and young branches on the outside of the trees as possible. Older foliage towards the inside of larger trees is frequently spotted by other pathogens or covered with sooty mold and should not be inspected. If symptoms are found on a tree, samples should be cut from those areas of suspect trees that show the best symptoms and should follow the sampling procedures above.

Nursery Survey Procedures: Citrus nurseries generally employ one, or a combination, of the following operational procedures: 1) trees grown outdoors in rows in the ground, 2) trees grown outdoors in containers (may be under shade cloth or in a lath house), 3) trees grown in a fully-enclosed screenhouse, generally in containers, and 4) trees grown in a greenhouse, generally in containers. Trees lined out in rows in the ground are usually budded sequentially by budders moving down the rows, whereas this may not be true of container-grown trees, which are moved about as nursery operations dictate. Nursery stock may be widely disseminated throughout a large geographic area, especially stock purchased by big retail chains, such as Home Depot and Lowe's, making it very important to thoroughly inspect all trees being grown in the nursery.

Depending on tree and row spacing, it may be necessary to walk every row in order to see all of the foliage on each tree. Examine newer foliage and branches on larger trees. Any budwood source trees located at the nursery should also be inspected. Use sampling procedures as indicated above. Mark the samples and the package to indicate that they are nursery samples. A hold will be placed on the nursery until the samples are processed; it is important to clearly mark the package so that the laboratory can give these samples priority

handling. Call the laboratory and advise them that nursery samples are being shipped. Several suspect trees sequentially occurring in a row of young, field-grown stock suggests the possibility that they were propagated from infected budwood. Question nursery personnel to determine the source of the budwood used to bud the stock. It may have come from a mature tree, or possibly budwood was cut from the tops of 1 to 2 year old trees previously propagated at the nursery. Inspect budwood source trees if available.

Sampling precautions: Before starting inspections, always determine if there have been recent pesticide applications that would make it unsafe to inspect the citrus trees. Check with property owners or managers for this information. Look for posted signs indicating recent pesticide applications, particularly in commercial groves. Determine if any quarantines are in effect for the area being surveyed, such as for root weevils, citrus canker, etc. Comply with all quarantine requirements. Before entering a new property, make certain that footwear is clean and free of soil to avoid moving soil-borne pests from one property to another.

If infected trees are found, the general recommendation is to remove all citrus within a 125 foot radius of infected trees and to apply an insecticide treatment for vector control to all trees within a 200 foot radius of the infected tree. These applications should be made according to the label directions until removal and disposal of infected and suspect trees is completed.

Key Diagnostics

Diagnosis of CVC in the field can be confused with other decline diseases of citrus, including citrus blight. CVC infected trees will take up water by syringe injection while blight infected trees do not. Diagnostic field symptoms are the small fruit having high sugar contents, the gummy lesion on the underside of the leaves, and small, pointy leaves at the top of the tree.

Classical procedures for diagnosis of *X. fastidiosa* have been based on cultural characteristics, serology, electron microscopy and molecular techniques. Recently, a number of polymerase chain reaction (PCR) procedures have been developed for the detection of *X. fastidiosa*. Primers have been developed that detect most, if not all, strains of *X. fastidiosa* (Firrao and Bazzi, 1994; Minsavage et al., 1994) that are specific to the strain that causes CVC (Pooler and Hartung, 1995; Beretta et al, 1997).

Fungal Diseases

Elsinoe australis

Scientific Name
Teleomorph: *Elsinoë australis* Bitancourt & Jenkins
Anamorph: *Sphaceloma australis* Bitancourt & Jenkins

Common Name(s)
Sweet orange scab, sarna o roña del naranjo dulce, roña de la naranja dulce, costra de los agrios sarna de la naranja dulce, antracnosis del naranjo, gale des agrumes, anthracnose de l'oranger.

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Ascomycota, **Class:** Loculoascomycetes, **Order:** Dothideales, **Family:** Elsinoaceae

Reason for inclusion in manual
National pest list and Regulated plant pest list

Pest Description
Two scab diseases are recognized on citrus, sweet orange scab caused by *Elsinoe australis* and citrus scab caused by *Elsinoe fawcettii* (Timmer, 2000). The teleomorphs of these fungi are from Brazil, and are morphologically similar. Ascospores of *E. ausytralis* are hyaline, straight or curved, 1 to 3 septate, slightly constricted at the septa, and the upper middle cell may be longitudinally septate (Sivanesan and Critchett, 1974). Most important, ascospores of *E. australis* (12 to 20 x 15to 30 µm) are bigger than those of *E. fawcettii* (5 to 6 x 10 to 12 µm) (Timmer, 2000). Their corresponding anamorphs, *Sphaceloma australis* and *Sphaceloma fawcettii* form nonseptate hyaline conidia (Sivanesan and Critchett, 1974), which are practically identical. *Elsinoe fawcettii*, however, forms colored, spindle-shaped conidia on scab lesions, whereas *E. australis* does not (Timmer 2000). Although, in culture, colonies of *E. australis* tend to be darker, and vinaceous to black, and those of *E. fawcettii* are usually beige to tan or brown, color might not be a distinguishing character (Tan et al., 1996; Timmer, 2000).

Little is known about the biology, ecology and epidemiology of *E. australis*. Available information is primarily based on studies with its anamorph *S. australis*. In culture, *E. australis* has an optimum growth temperature of 24 to 29 ºC (Knorr, 1863; Whiteside, 1975). Since *E. australis* does not infect leaves, its survival during the absence of fruits is a

mystery; perhaps the sexual stage plays a greater role in the survival and dispersal of the pathogen (Timmer 2000). It is also possible that insects or wind-carried water droplets containing spores may contribute to the spread of the pathogen. *Elsinoe australis* only attack young host tissue.

Pest importance

Elsinoe australis, the sweet orange scab pathogen, is economically important because it attacks citrus species grown for the fresh-fruit market (oranges and mandarins). It is also important because the pathogen can be carried on fruits in international trade.

Sweet orange scab is widespread in wet subtropical and cooler tropics of South America. Knorr (1963) reported that in some Brazilian groves, 50 to 60% of the orange crop were found to be blemished by sweet orange scab, and in other groves, 1/3 of the crop had to be culled before shipment.

Figure 1. Symptoms of sweet orange scab on sweet orange in Brazil. Photo Courtesy of Eduardo Feichtenberger.

Symptoms

Fruits are infected in the early stages of their development, and infections do not extend into the albedo. Initially, scab pustules (mixture of fungal and host tissue) are slightly raised and pink to light brown (Fig. 1). Later, they become warty, cracked, yellow-brown, and eventually dark grey (Timmer, 2000). Lesions are 2 to 6 mm in diameter and may deform the rind, but the normal round shape returns as fruits mature (Knorr, 1963). In contrast to the more common citrus scab, lesions of sweet orange scab on oranges and tangerines are flatter (Timmer, 2000; Bitancourt & Jenkins, 1937), nearly rounded and less wrinkled (Jenkins 1931, Bitancourt & Jenkins, 1937). Because symptoms of citrus scab vary with cultivar and tissue, differentiation of these two diseases is difficult when based only on symptoms (Timmer, 2000).

Known Hosts

Elsinoe australis is known to primarily attack sweet orange and tangerines. It is most important on navel orange (*Citrus sinensis*) and mandarin (*C. reticulata*) fruit. It also affects lemons (*C. limon*), satsumas (*C. unshiu*), limes (*C. aurantifolia*), oval kumquat (*Fortunella margarita*), and fruit of many other citrus species (Timmer, 1996).

Known Distribution

Sweet orange scab is largely confined to the humid citrus producing areas in southern South America (Timmer, 2000). It has been found in Argentina, Bolivia, Brazil, and Paraguay (Sivanesan and Critchett, 1974). Its presence in Uruguay has not been confirmed (Diaz *et al*., 1992). Its presence in South America can be traced back to as early as 1882

(Jenkins et al., 1933). It was also reported in Oceania from Cook Island, Fiji, Niue, and Samoa (CABI, 2004; Sivanesan & Critchett, 1974).

Potential distribution within the US

To date, no records exist regarding the presence of *E. australis* in the US. If introduced, it might become established in humid and warm areas or in areas where new flush and fruit setting coincide with spells of relatively warm and humid weather. Citrus growing areas with less than 1300 mm of annual rainfall, extended hot seasons (> 24°C), or dry summers are not favorable for sweet orange scab (e.g., California and Arizona).

Survey/Key Diagnostics

Currently, there are no specific published survey methods for this disease. *Elsinoe australis* can only attack fruit and there is no evidence that the pathogen is capable of infecting leaves of any citrus species (Timmer et al., 1996; Timmer, 2000). Surveys based on fruit symptomatology could yield either of the two scab diseases recognized on citrus and caused by *Elsinoe australis* and *Elsinoe fawcettii*; however, *E. australis* forms larger, smoother, more circular scabs, whereas *E. fawcettii* scabs are typically irregular, warty, with deep fissures. *Elsinoe australis* does not produce in the scab lesions the colored, spindle-shaped conidia that *E. fawcettii* produces.

Elsinoe australis can be differentiated from other scab pathogens by its morphology, tissues attacked, host range (Timmer *et al*., 1996), and the use of molecular techniques (Tan *et al*., 1996; Hyun, *et al*., 2001)

Elsinoe australis can be isolated by surface sterilizing symptomatic fruit for 1 minute in 75% ethanol, then for 1 minute in 1% sodium hypochlorite, followed by rinsing in sterile distilled water and drying. Thereafter, scab pustules are scraped with a scalpel blade to deposit flakes of diseased tissue on ½ strength PDA, or PDA containing 400 µg a.i. /ml of Dodine® (Hyun et al., 2001). To produce conidia, 5 to 10 mm^3 pieces of mycelium are removed from 2 to 3 week-old cultures on PDA and crushed in a plastic Petri plate with a spatula. Resulting fragments are stirred into liquid modified Fries medium, and incubated for 2 days at 27°C. Plates are washed three times with sterile distilled water, flooded with autoclaved farm pond water, placed under long-wavelength (366 mm) UV light for 1 hour, and incubated overnight in the dark at 20 to 25 °C. (Timmer et al., 1996).

In contrast to the cumbersome classical identification of *E. australis*, this pathogen can be detected by using molecular markers. Tan *et al*. (1996) differentiated *E. australis* from other scab pathogens by using restriction analysis of the amplified ITS of ribosomal DNA with several endonucleases. *Elsinoe australis* was also genetically differentiated by using random amplified polymorphic DNA markers (Tan, *et al*., 1996; Hyun *et al*., 2001).

Guignardia citricarpa

Scientific Name
Teleomorph: *Guignardia citricarpa* Kiely
Anamorph: *Phyllosticta citricarpa* (McAlpine) Van der Aa

Synonyms:
Phoma citricarpa, Phyllostictina citricarpa

Common Name(s)
Citrus black spot.

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Ascomycota, **Class:** Loculoascomycetes, **Order:** Dothideales, **Family:** Botryosphaeriaceae

Reason for inclusion in manual
National pest list and Eastern region pest list

Pest Description
Two strains of *G. citricarpa* were recognized in the past. Of the two, one is pathogenic to citrus and the other is non-pathogenic, but widespread on other hosts and geographically distributed (McOnie, 1964). Baayen et al. (2002) designated the widespread non-pathogenic strain as *G. mangiferae* (*anamorph: Phyllosticta capitalensis*) and the citrus black spot pathogen as *G. citricarpa*.

According to Sutton & Waterston (1966) and van der Aa (1973), as revised and amended by Baayen et al. (2002), perithecia of *G. citricarpa* are formed exclusively on fallen decomposing leaves, not in fruit, on leaf lesions or on media. Perithecia are solitary or aggregated, globose to pyriform, dark brown to black, 125 to 360 µm in diameter, ostiole papillate, circular, 10 to 17.5 µm in diameter, with no paraphysis and periphysis. Asci are clavate cylindrical, shortly stipitate, 40 to 65 x 12 to 15µm, bitunicate, with eight spores each, and ascus wall thick. Ascospores are hyaline, nonseptate, multiguttulate and swollen in the center, 4.5 to 6.5 x 12.5 to 16 µm, and colorless appendage at each end. If perithecia form in culture they are usually infertile (Kotzé, 2000).

Pycnidia are found in abundance on dead fallen leaves and fruits, usually developing in culture. They are mid to dark brown, solitary, sometimes aggregated, globose, immersed, 70 to 330 µm in diameter, sclerotioid on the outside, pseudoparenchymatous within, ostiole darker, slightly papillate, circular and 10 to 15µm diameter. Conidia are obovate to elliptical, hyaline, nonseptate, multiguttulate, apex slightly flattened with a colorless subulate appendage, base truncate, 5.0 to 8.5 x 9.4 to 12.7 µm, surrounded by a barely visible colorless gelatinous coat (<1.5 µm thick). Conidia and the conidial sheath are best examined using a microscope equipped with differential interference contrast or using

Indian Black ink, which will make the sheath appear translucent against the black background.

Biology and Ecology

Guignardia citricarpa can be present for several years in a particular area before any symptoms are noticed; it may take 5 to 30 years from its first detection to reach epidemic levels (Kotzé, 1981). The most important source of inoculum of *G. citricarpa* is the airborne ascospores (Kotzé, 1981; McOnie, 1967). Pseudothecia develop in decomposing leaves after 40 to 180 days of leaf fall, depending on frequency of wetting and drying. The optimum temperature for ascomata formation is 21 to 28°C. Once ascospores mature, rainfall or irrigation may trigger their release. Ascospores are carried by wind throughout the canopy and may reach long distances.

When ascospores land on fruit or vegetative tissues under moist conditions, they germinate and form an appressorium. The infection peg then penetrates the cuticle and epidermis to form latent infections on fruit or leaves. These infections will develop later and produce the typical symptoms once the fruit attains full size or reaches maturity. *Guignardia citricarpa* seldom infects leaves, but the fungus colonizes the dead leaves as a saprophyte and later forms either pycnidia or pseudothecia.The anamorph probably plays only a minor role in the disease cycle. Conidia produced on dead leaves can only reach fruit and leaves by splash dispersal, whereas, conidia produced on fruit can be washed down through the canopy and infect leaves and young fruits (Kotzé, 2000). Fruits are susceptible for at least 4 to 6 months after petal fall (Baldassari, 2001); thus, ascospores must be present during this period. High temperatures, high light intensity, drought, and poor tree vigor may accelerate disease development.

Figure 1. Symptoms of hard spot or shot-hole type of black spot. Photo Courtesy of N. A. Peres, Univ. Florida, GCREC. Photo insert Courtesy of Renato Ferrari dos Reis.

Pest importance

Fruit with necrotic lesions are unacceptable for export, thus, most losses are caused by the presence of these external blemishes. Losses due to fruit drop and costs for chemical control may also occur in years favorable to *G. citricarpa*, or when fruit is held on the trees past peak maturity.

Symptoms

Several types of symptoms occur on citrus and often overlap (hard or shot-hole spot, freckle spot, virulent spot, and false melanose). Hard spot/shot-hole spot lesions (Fig. 1, and Fig. 2A) are most common and usually appear after the fruit has started to turn yellow or orange. They are typically crater-like lesions with a light center, dark-brown to black rim, often with a green halo on mature orange fruit, and 3 to 10 mm in diameter. Pycnidia are often present in these lesions as tiny black dots inside of the rim. Freckle spot/speckled blotch are grey, tan, reddish, brownish or not discolored at all, slightly depressed, 1 to 3 mm in diameter and occur late in the season after harvest (Fig. 2B). Pycnidia may be incidentally present. Freckle spots often occur as satellite spots around hard-spot lesions. Many intermediate types can also occur between freckle and hard-type lesions. Von vrulent lesions, spots may coalesce and form large, slightly sunken brown to black areas on mature fruit. This type is the most damaging, because lesions can extend deeply into the rind, develop a leathery texture, and cover the entire fruit (Fig. 2C).

Figure 2. Symptoms of hard spot type of black spot on Valencia fruits (A), Freckle or Cracked spot type of black spot (B), Symptom of virulent spot type of black spot (C), Symptom of false melanose type of black spot (D). Photo A courtesy of Renato Ferrari dos Reis. Photos B-D courtesy of N. A. Peres, Univ. Florida, GCREC.

False melanose usually appears on green fruit as raised dark-brown to black specks that may coalesce (Fig. 2D). Pycnidia do not form on these lesions.

Lesions on leaves are rare on most citrus, but more frequent on lemons. They first appear in mature leaves as tiny circular red to red-brown spots (Fig. 3) that do not exceed 3 mm. Later they become sunken necrotic spots with a light center and dark brown or black ring, often with yellow halo.

Figure 3: Leaf with black spot. Photo Courtesy of N. A. Peres, Univ. Florida, GCREC.

Known Hosts

Citrus spp. All commercial cultivars are susceptible to black spot except the sour orange (*C. aurantium*) and its hybrids. Lemons are particularly susceptible to *G. citricarpa*.

Known Distribution

Guignardia citricarpa is present in China, Indonesia, Russia, South Africa (localized), Zambia, Zimbabwe,

Argentina (localized), Brazil (localized), and Australia (localized). Reports of its presence on citrus in other countries may be confused with the non-pathogenic species, *G*. *mangiferae* (CABI, 2005).

Potential distribution within the US

Climatic conditions in Florida are favorable for *G*. *citricarpa*, although this fungus has not yet been found there (Kotzé, 1981; Timmer, 2005).

Survey/Key Diagnostics

Currently, there are no specific survey methods for this disease. Black spot can be identified with considerable certainty if hard spot lesions with tiny black dots (pycnidia) are present on the fruit. Freckle spot, false melanose and virulent spot can easily be confused with other diseases and injuries (Kotzé, 2000). If black spot occurs in a new location, it will be seen first on lemons.

Black spot is almost exclusively a fruit disease, and primarily occurs on the side of the fruit exposed to sunlight. The key diagnostic symptom is the rounded dark brown lesions enclosing a depressed, light brown to gray-white center with several pycnidia.

Black spot symptoms appear during the later stages of fruit development or after picking. Incidence of disease is more evident in fruit in the upper canopy and more severe in older trees (Kotzé, 1981). Available inoculum on fallen leaves can be monitored in the laboratory (Truter et al., 2004).

Guignardia citricarpa is relatively easy to isolate from virulent spot or hard spot lesions. Isolation from other types of symptoms can be difficult or impossible. Isolation can be made by transferring small surface sterilized dissected tissue pieces on carrot dextrose agar, cherry agar, or PDA, and incubating it between a temperature range of 23 to 27ºC. These temperatures are optimum for sporulation and mycelial growth (Mendes et al., 2005). Possible colonies of *G*. *citricarpa* are slow-growing and dark in color, and should be transferred to cherry agar for growth-rate testing and on oat meal agar for pigment production. To induce sporulation, original cultures should be placed at 22ºC, under near-ultraviolet light. Pycnidia should form in 14 days on cherry agar.

The presence of the cosmopolitan *G*. *mangiferae* makes the identification of *G*. *citricarpa* difficult and time consuming. Baayen et al. (2001) reported that *G*. *citricarpa* grows slower on cherry decoction agar than *G*. *mangiferae*. Colonies of *G. citricarpa* are lighter, more lobate, and have a wider translucent zone than *G*. *mangiferae*. *Guignardia citricarpa* produces a yellow pigment on oatmeal agar that is not produced by *G*. *mangiferae*. *Guignardia mangiferae* usually produces fertile perithecia in culture in contrast to *G*. *citricarpa*. Conidia of *G*. *citricarpa* lack the visible mucoid sheath present on those of *G*. *mangiferae*.

Lesions on fruit can also be incubated to stimulate the formation of pycnidia. Fruits are disinfested in 1% sodium hypochlorite, washed thoroughly and dried. Lesions with no pycnidia are removed with a cork borer (7 mm diameter) or scalpel blade, and aseptically placed in Petri dishes with wet filter paper. The Petri dishes are incubated for five days

under continuous light at 27ºC; thereafter, lesions are examined for the presence of pycnidia, and slides are prepared to examine the conidia (OEPP/EPPO, 2003).

PCR methods may also be used to differentiate these two species (Tan et al, 1996). A primer pair has been developed by G. C. Carroll (University of Oregon) and P. G. M. Bonants (Plant Research International, Wageningen, Netherlands) to differentiate *G. citricarpa* from other species of *Guignardia* (see OEPP/EPPO, 2003).

Phoma tracheiphila

Scientific Name
Phoma tracheiphila (Petri) Kantschaveli and Gikachvili

Synonyms:
Deuterophoma tracheiphila, Bakerophoma tracheiphila

Common Name(s)
Mal Secco, citrus wilt

Type of Pest
Plant pathogenic fungus

Taxonomic Position
Phylum: Deuteromycota, **Class:** Coelomycetes, **Order:** Sphaeropsidales, **Family:** Pleosporaceae

Reason for inclusion in manual
National pest list

Pest Description
Mature black ostiolate pycnidia (60 to 165 x 45 to 140 µm diameter) bear a neck. Within the pycnidial cavity, minute unicellular, mononucleate and sometimes binucleate hyaline conidia (0.5 to 1.5 x 2 to 4 µm) are produced by conidiogenous cells (phialides). Sometimes conidia are extruded through ostioles in whitish cirri. Larger conidia (phialoconidia) are produced by phialides (12 to 30 x 3 to 6 µm) borne on free hyphae growing on exposed wood surfaces, wounded plant tissues, and within the xylem elements of the infected host; they are hyaline, unicellular, uninucleate and sometimes binucleate or trinucleate, straight or curved, with rounded apices (3 to 8 x 1.5 to 3 µm).

No teleomorph (sexual stage) is known. For more information see Graniti (1955). A full description is given by Ciccarone (1971) and by Punithalingam & Holliday (1973).

Germination of conidia and host penetration require around 40 hours of moisture at a temperature of 15 to 16 °C. After infection, the fungus progresses to the lumen of the xylem vessels then spreads systemically, and mainly, upward. Systemic movement is assisted by phialoconidia that are translocated in the xylem sap (Solel and Salerno, 1989). Infection may occur via roots, particularly if damaged through cultivation. The range of temperature at which infection will occur is considered to be between 14 and

Figure 1. Lemon tree severely infected with Mal Secco disease. Photo courtesy of CABI, 2004.

28°C. The optimum temperature for growth of the pathogen and for symptom expression is 20 to 25°C, whereas the maximum temperature for mycelial growth is 30°C. The length of the incubation period may vary according to the seasons. Prunings containing affected twigs or branches can be a source of inoculum for several weeks. The fungus can survive within infected twigs in the soil for more than four months.

Pycnidia in infected tissues produce numerous conidia that can be dispersed by water. Conidia are locally dispersed by raindrops falling on the inoculum source and by wind-blown rain (Solel and Salerno, 1989). Long range dispersal can occur via the movement of diseased propagation material. Infection typically occurs through wounds, such as those caused by wind and hail storms, frost, and during pruning and harvesting operations.

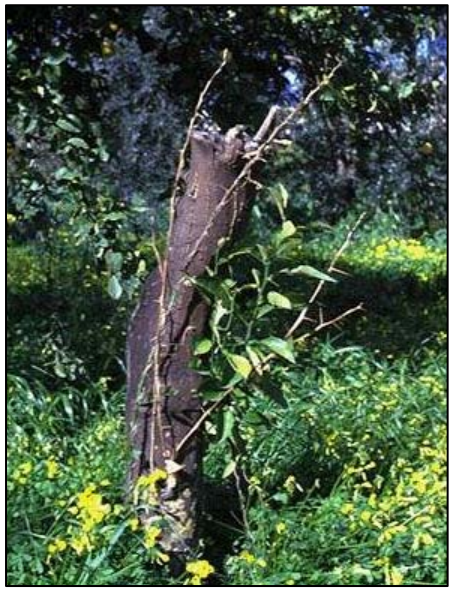


Figure 2. Suckers (sprouts) from the base of rootstocks. Photo courtesy of CABI, 2004.

Pest Importance

Mal Secco, Italian for dry disease, was first reported on the Greek island of Chios in 1894 (Fig. 1). The disease is apparently confined to the citrus-producing areas of the Mediterranean Basin, around the Black Sea and Asia Minor. In the Mediterranean region, *P. tracheiphila* is the most destructive fungal disease of lemons. Reduction in lemon yield in Italy has resulted in an estimate of more than US$160 million in annual losses. Up to 100% of trees in a lemon orchard of a susceptible cultivar can be affected. Destructive outbreaks of *P. tracheiphila* may occur after frost spells and hail damage in spring. In general, injury to the tree through severe cold weather may predispose it to fungal attack. The symptoms of the disease are most severe in spring and autumn. During high summer temperatures, the spread of the pathogen in the host vascular system ceases and symptoms do not further develop. The disease has reduced the quantity and quality of lemon production in the areas where the pathogen is present, and limits the use of susceptible species and cultivars (CABI, 2004).The fungus kills nearly one million trees yearly. Because the disease not only lowers production, it also kills trees and adds to the seriousness of the disease

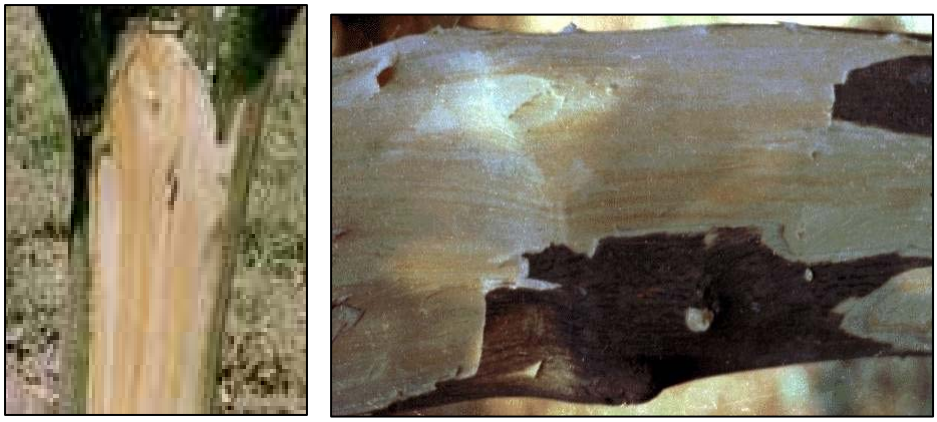


Figure 3. Diagnostic salmon pink to reddish orange coloration of the wood. Photos courtesy of CABI, 2004.

Symptoms

Trees are susceptible to infection at any age, but young trees tend to more severely infected. Symptoms depend upon the mode of infection. When infection starts in the canopy, the first symptoms appear in spring as leaf (veinal) and shoot chlorosis; as the disease progresses, the leaves wilt, dry up, fall and dieback occurs. Sometimes fallen leaves show a red discoloration in the midrib and some secondary veins. In these cases, the pathogen slowly spreads downward from young shoots to branches and main limbs, then to the trunk and roots. Infected bark, particularly on shoots of one to two years old, may become lead-grey to silver-grey. Eventually, the bark ruptures to reveal numerous raised black pycnidia beneath the surface. When infections occur at the base of the trunk or in the roots, the pathogen rapidly progresses upward. Symptoms may be produced over the whole tree, or in only one limb. The disease may develop so suddenly that the leaves dry up on the tree. In one distinctive type of root infection, the pathogen invades the inner xylem vessels. In these cases there are no obvious symptoms at first, but eventually the pathogen reaches the outer wood and causes a sudden collapse of the canopy. When infection starts in the canopy, it may take several years for the disease to move downward and involve the trunk; however, when the disease originates as a basal or root infection, tree collapse can be rapid.

Figure 4. Browning of the heartwood caused by 'mal nero'. Photo courtesy of CABI, 2004.

The growth of sprouts from the base of the affected branches and suckers from the rootstock are a common response of the host to the disease (Fig. 2). On cutting into the infected twigs or peeling back the bark, the characteristic salmon-pink or orange-reddish discoloration of the wood can be seen (Fig. 3); this internal symptom is associated with gum production within the xylem vessels.

In addition to the more common form of mal secco, two different forms of the disease can be distinguished: "mal fulminante," which is a rapid and fatal form of the disease due to root infection; and "mal nero," which is a consequence of chronic infection of the tree leading to a browning of the heartwood (Fig. 4).

When unripe lemon fruit is infected, it shows partial or total yellowing of the peel, depending on the age of the infection. When ripe lemon fruit is infected, it turns dark yellow, almost reddish, in color (Ippolito *et al*., 1987).

Known Hosts

Primary hosts are *Citrus,* particularly *C. bergamia* (bergamot)*, C. aurantiifolia* (lime)*, C. aurantium* (sour orange)*, C.limon* (lemon)*,* mandarin (*Citrus reticulata*) and *C. medica* (citron). Other hosts include *Poncirus*, *Severinia,* and *Fortunella* spp. The disease is most prevalent and severe on lemon and citron. Bergamot, some mandarins, tangelos, and tangors are also vulnerable; however, grapefruit and sweet orange are rarely infected and usually display less severe symptoms. Affected rootstocks include rough lemon, limetta, alemow, sour orange, and Troyer and Carrizo citranges. (CABI, 2004)

Known Distribution

Mal Secco occurs in countries around the Mediterranean Basin, Black Sea and Asia Minor. Current distribution incudes Algeria, Tunisia, Cyprus, Iraq, Israel, Lebanon, Syria, Turkey, Yemen, Albania, France, Greece, Italy, Mediterranean countries, and the Russian Federation (CABI, 2004).

Potential Distribution within the US

Phoma tracheiphila has a wide range of natural hosts in the family Rutaceae, including *Citrus* spp., *Poncirus trifoliata*, *Fortunella* spp. and *Severinia buxifolia.* Lemon, citron, and mandarin and some of its hybrids are highly susceptible to the disease. Lime has been severely affected in Israel.

The temperature range at which infection occurs is between 14 and 28°C. The optimum temperature for growth of the pathogen, and for symptom expression, is 20 to 25°C. The maximum temperature for mycelial growth is 30°C. Susceptible hosts and environmental conditions conducive to infection occur within the US.

Survey

Movement of infected plant parts of *Citrus spp.* and other rutaceous hosts could introduce the pathogen into new areas. Infected propagative material would be the most likely means of spread. This material could easily serve as a source of inoculum due to conidium production in and on infected plant parts. The likelihood of infected fruit serving as a source of disease spread is very low; they are of such poor quality that they are unlikely to be marketed or exported. Leaves become infected, but are chlorotic and dry, they are unlikely to be moved commercially for consumption.

Phoma tracheiphila has been detected in lemon seeds (Stepanov and Shaluishkina, 1952). *Phoma tracheiphila* survives in lemon seed as mycelium, but is unable to pass from the seed coat to developing seedlings (Ippolito *et al*., 1987). There is no evidence that it is seedborne on other seed types.

To detect Mal Secco in the field, look for leaf and shoot chlorosis, wilt, and defoliation, followed by dieback of twigs and branches. Upper canopy infections spread slowly, but rapid wilting and death of tree portions may occur as a result of infection of lower portions of the tree or at the base of large branches. On live twigs and branches, make diagonal cuts and look for the diagnostic orange-yellow to pink-salmon discoloration that develops in newly infected twigs and branches. Pigmentation can be accentuated by wetting the cut surface of the wood with a 1% solution of potassium hydroxide or ammonium hydroxide. The discoloration becomes brownish or black in later stages of the disease. On dead twigs and branches, look for bark that has become gray. Examine microscopically for embedded pycnidia containing minute, colorless conidia. When submitting material for identification or confirmation of *P. tracheiphila*, discolored twigs and branches, or bark with pycnidia, will be most useful for identification purposes. Suspect plant material should be dried, labeled, and placed in sealed double containers.

Acervuli of the *Colletotrichum* state of *Glomerella cingulata*, a secondary invader of withered twigs, are often associated with the pycnidia of *P. tracheiphila*.

Key Diagnostics

Mal Secco is recognized in the field by the above symptoms. A serological test (ELISA) and a polymerase chain reaction technique have been developed for detecting Mal Secco (Nachmias, 1979, Rollo et al., 1990). The disease can be difficult to detect in propagation material. It may be present, without visible symptoms, in roots, fruits, seeds, and possibly in wood (Plant Health Australia, 2002).

A reliable diagnostic sign of Mal Secco is the red or orange coloration that appears in the recently invaded xylem. This can be revealed by either peeling off the bark or cutting through the wood. Symptoms can be enhanced by applying an ammonium or a potassium hydroxide solution. The fungus can be easily isolated from infected xylem on agar medium (USDA, 1984).

Viral Diseases

Citrus leprosis virus

Scientific Name
Citrus leprosis virus (acronym: CiLV)

Common Name(s)
Citrus leprosis, leprosies

Type of Pest
Plant pathogenic virus

Reason for inclusion in manual
Sometimes mistaken for citrus canker (*Xanthomonas axonopodis* pv. *citri)* lesions and for citrus psorosis virus

Pest Description
The bulk of available taxonomic evidence suggests that *Citrus leprosis virus* is the causal agent of citrus leprosis. This virus is an unassigned species of the family Rhabdoviridae.

Two different virus particles are associated with leprosis symptoms, one of nuclear type and another of cytoplasmatic type (Colariccio et al., 1995). The presumed virus particles occur in parenchyma cells of the infected orange leaves, fruits or stems. Particles are short, bacilliform, 120 to 130 nm long (occasionally up to 300 nm) and 50 to 55 nm wide (Fig. 1). They occur within the lumen of the endoplasmic reticulum. In addition to the presence of the rhabdovirus-like particles within the endoplasmic reticulum of tissues from the lesion, dense viroplasm-like material is commonly found in the cytoplasm, near the particles. Small vesicle-containing fibrillar materials are frequently present in the vacuole associated with the tonoplast next to the dense material (Colariccio et al., 1995, Lovisolo, et al., 1996).

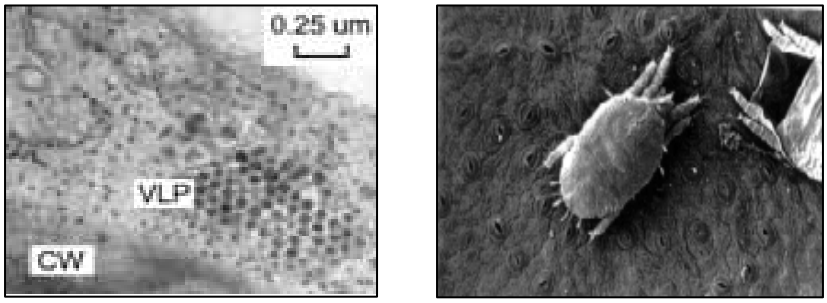


Figure 1. Transmission electron micrograph of *Citrus leprosis virus* particles (VLP) in leaf of sweet orange. (CW = cell wall.) (left) and scanning electron micrograph of *Brevipalpus phoenicis* vector of *Citrus leprosis virus* (right). Photos Courtesy of Jos Carlos Verle Rodrigues and Marinês Bastianel

Citrus leprosis virus is transmitted by false spider mites or flat mites (Fig. 1). So far, only the cytoplasmic type has been experimentally transmitted by *Brevipalpus* mites. Seedlings of Cleopatra mandarin and Seleta or Pêra sweet oranges infested with viruliferous mites (*B*.

phoenicis) develop symptoms of leprosis in 1 to 2 months (Rodrigues, et al., 2000). Because of its non-systemic infection, leprosis can only be important where attacks by its vector mites are significant. Species of *Brevipalpus* involved as vectors of CiLV include *B. phoenicis*, *B. californicus*, and *B. obovatus*. These species have been identified from citrus in Brazil, Costa Rica, Honduras, South Africa, Florida and Texas. *Brevipalpus phoenicis* is recognized as the vector of citrus leprosis in Brazil; *B. obovatus* serves as a vector in Argentina and Venezuela (Rodriguez et al., 2003). *Brevipalpus californicus* was the reported vector of leprosis in Florida (Knorr et al., 1968). Mites that acquire CiLV can transmit the virus throughout their life, even if they are maintained on unsusceptible plants; individuals from eggs of viruliferous females cannot transmit the virus.

Brevipalpus mites can inject toxic saliva into fruits, leaves, stems, twigs, and the bud tissues of numerous host plants, including citrus (Childers, et al, 2003a). Severe infestation of mites in citrus trees can cause leaf drop and the downgrading of fruit. In Texas, *B. phoenicis* and *B. californicus* cause more damage to fruit in inside branches and in the lower canopy (< 2 m) (French and Rakha, 1994). Lesions on fruit surfaces are initially depressed yellowish circular areas, gradually developing a brown necrotic area or spot in the center (3 to 12 mm diameter) that can become darker and corky in texture. Lesions may coalesce, cover large areas (50 to 75 mm) and display a distinct raised surface on the fruit. On Marrs, Ambersweet, and navel oranges, the brownish irregular-shaped spots (1 to 30 mm) occur more often on the styler end of the fruit and in the inner canopy (Childer, et al., 2003a). Under controlled conditions, *B. californicus* fed on the twigs, petioles, upper and lower leaf surfaces and developing buds of *Citrus aurantium* seedlings. As a result of feeding injury, leaves were rounded and severely stunted. Leaves also had marginal necrosis and tip burn. Mite feeding resulted in severe stunting of new shoots and the formation of corky swollen buds (Childers, et al., 2003a).

Figure 2. Symptoms of citrus leprosis on leaves and in fruits of Pêra sweet orange. Photo courtesy of Marinês Bastianel.

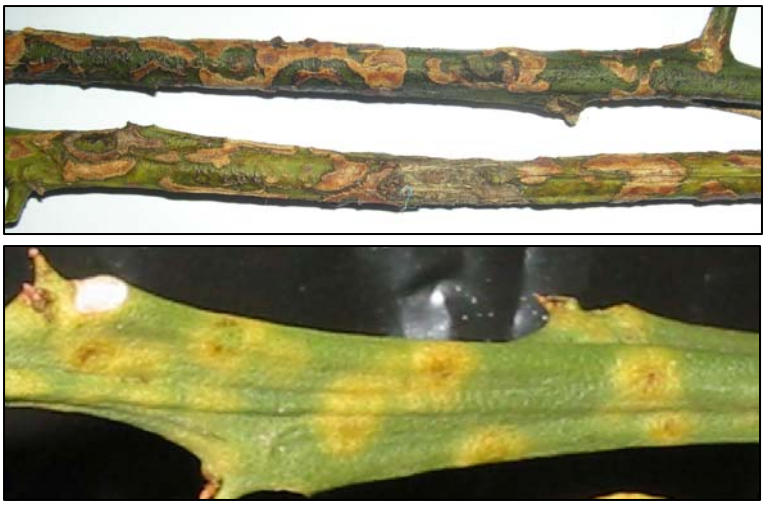


Figure 3. Symptoms of citrus leprosis on branches of Pêra sweet orange. Photo courtesy of Marinês Bastianel.

Pest Importance
Citrus leprosis nearly destroyed the citrus industry in Florida between 1906 and 1925 (Knorr, 1968). The disease is no longer present in Florida nor does it occur in Texas (Childers, et al., 2003b). Recently, it has been found in Panama (Dominguez, et. Al., 2000), Costa Rica, and Guatemala; there are fears that it can/will spread throughout Central America and eventually reach the US. Citrus production in the US is a billion dollar industry. If CiLV invades and becomes established in the U.S., it may cause serious economic damage in all citrus-growing states. In Brazil, where citrus leprosis occurs in all major citrus areas, the estimated annual cost of keeping mite populations under control is more than $100 million dollars (Rodriguez et al., 2003).

Symptoms
The viruses transmitted by *Brevipalpus* cause symptoms on leaves, fruits, and twigs of host plants. Each lesion is caused by a feeding puncture of a virus-infected mite; however, symptoms often do not appear, and the virus remains latent. On leaves (Fig.2), lesions are usually round, with dark-brown central spots (2 to 3 mm in diameter). These lesions are surrounded by a chlorotic halo in which 1 to 3 brownish rings often appear surrounding the central spot. Lesion size may vary from 10 to 30 mm, but they may coalesce to form large lesions. On green fruits, the lesions are initially yellowish, later brownish or blackish (10 to 20 mm in diameter), sometimes depressed, and occasionally with gum exudation (Fig. 2). On stems, lesions appear as grey or brown cortical bark scaling (Fig. 3). Lesions may coalesce and cause the death of the twig (Rodrigues et al., 2003; Lovisolo, 2001). Heavy infections, as described in 'lepra explosiva' in Argentina, may result in severe defoliation, fruit fall, and eventually tree death.

Known Hosts
According to Lovisolo (2001), natural hosts of CiLV include Citrange (*Citrus sinensis* x *Poncirus trifoliata*), citron (*C. medica*), Cleopatra mandarin (*C. reshni*), grapefruit (*C. paradise*), lemon (*C. limon*), mandarin (*C. reticulata*), Mexican lime (*C. aurantifolia*), Persian lime (*C. latifolia*), rough lemon (*C. jambhiri*), sour orange (*C. aurantium*), sweet orange (*C. sinensis*), and tangor (*C. reticulata* x *C. sinensis*). No other plant species is known to serve as a natural host. Experimental hosts include *Atriplex hortensis*, *Atriplex latifolia*, *Beta vulgaris* subsp. *cicla*, *Chenopodium album*, *Chenopodium amaranticolor*, *Chenopodium bonus-henricus*, *Chenopodium capitatum*, *Chenopodium foliosum*, *Chenopodium murale*, *Chenopodium polyspermum*, *Chenopodium quinoa*, *Gomphrena globosa*, and *Tetragonia tetragonioides*.

Known Distribution
Citrus leprosis has been found in South America: Argentina, Bolivia, Brazil, Paraguay, Uruguay, and Venezuela (Gomez, et al., 2005; Kitajima et. al., 1974; Rodriguez et al., 2003), and Central America: Costa Rica, Guatemala, and Panama (Dominguez, et. al., 2000). Reports of its presence in other countries have not been confirmed.

Potential Distribution within the US
Citrus leprosis is currently not present in the US (Childers, et al. 2003); however, there have been reports of it occurring in Florida between 1905 to 1925 (Knorr et. al., 1968). If

introduced, it may become established in all citrus growing areas in the US. Its vectors, the flat mites (*B*. *phoenicis*, *B*. *obovatus* and *B*. *californicus*), are already widely distributed and abundant in California, Florida and Texas (Childers et al., 2003a; French and Rakha, 1994; Knorr et al., 1968).

Survey

Traditionally, citrus leprosis has been detected by the examination of localized lesions on leaves, fruits, twigs, or the presence of flat mites (of the genus *Brevipalpus)* on lesions. The examination of symptoms is not a reliable method of detection because of the similarity between leprosis and other citrus diseases, such as citrus canker, psorosis, and zonate chlorosis. Zonate chlorosis is a disease of unknown etiology that is associated with infestations of *B*. *phoenicis*; it is characterized by the presence of concentric green and chlorotic rings, but no necrosis. Bark scaling in leprosis is more eruptive than in psorosis.

Key Diagnostics

Citrus leprosis virus can be detected by inoculating extracts of suspected leprosis-infected leaves, fruits, or stems to diagnostic plants. Lesions on *Chenopodium* species are brown with a clear center, chlorotic halo, and are 1.5 mm in diameter. Lesions in *Gomphrena globosa* are similar, but reddish-brown in color. Sweet orange cv. Caipira shows faint, but clear brown rings, about 2 mm in diameter, with clear centers, and chlorotic halos. In *C*. *quinoa* and *C*. *amaranticolor*, local lesions appear in 5 to 7 days; in *G. globosa* in 14 to 15 days; and in sweet orange cv Caipira in 24 days. Best results are obtained if test plants are grown in growth chambers at 30 to 32ºC (day) and 24 to 25ºC (night), and if older leaves are used for inoculations (Colariccio, et al., 1995; Lovisolo, et al., 1996).

Until recently, the only diagnostic method of observing the CiLV particles in diseased tissue was by transmission electron microscopy, which is time consuming, sample limiting, and expensive. Current advances in molecular technology, such as the RT-PCR technique, have allowed the development of specific primers for CiLV detection (Locali, et al., 2003). Locali et al. (2003) have shown high specificity for CiLV using two specific primer pairs designed from amplified CiLV cDNA that was cloned and sequenced. The high specificity of the primer pairs was evident when tested against other viruses, such as CTV. This RT-PCR molecular diagnostic tool may help to diagnose the disease faster and more accurately. Gomez et al. (2005) used this technique to detect CiLV in Bolivia.

Citrus psorosis virus

Scientific Name
Citrus psorosis virus (acronym: CPsV)

Common Name(s)
Psorosis, psorosis of citrus, scaly bark, citrus scaly bark, scaly bark of citrus.

Type of Pest
Plant pathogenic virus

Reason for inclusion in manual
Western Region pest list (Hawaii)

Pest Description
Virus particles associated with psorosis (CPsV) were first described by Derrick et al. (1988). CPsV is a multi-component single-stranded RNA virus with a coat protein whose molecular mass is approximately 48 kDa (Derrick and Barthe, 2000). CPsV particles are highly kinked, thread-like, spiral filaments, which occur in various configurations of two distinguishable sizes. The short particles are about 690 to 760 nm in length, and the larger particles four times longer (Fig. 1). Several strains of CPsV have been detected by biological, serological and molecular techniques (Derrick and Barthe, 2000).

CPsV is not restricted to the phloem. Tissue immunoprints show that it also invades parenchyma cells (Martín et al., 2002). In most citrus areas, psorosis appears to spread only through propagation of infected buds. There are reports of natural dispersal of psorosis-like bark scaling in Argentina, Uruguay and Texas; however, the presence of CPsV in these plants was not confirmed, thus, no natural vector is known (Milne et al., 2003).

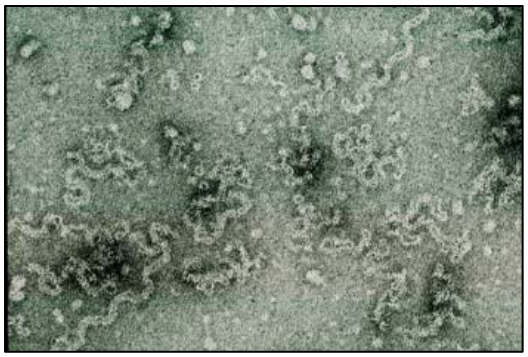


Figure. 1. Electron microscope photo of the long form particles of CPsV. Photo Courtesy of R. G. Milne

Experimentally, the CPsV virus is readily transmitted from citrus to citrus by grafting. It remains in the callus grown from infected plants and can be transmitted by grafting these callus cells under a bark flap of the receptor plant (Navas-Castillo et al., 1995). Seed transmission was reported, but adequate biological indexing was not done. Recent tests of seed transmission have yielded negative results (Roistacher, 1993; D'Onghia et al., 2000).

Pest Importance

Psorosis is still a serious problem in certain citrus growing areas of the world. It has been eliminated in most areas by using clean budwood. The bark-scaling forms of psorosis are destructive and cause the debilitation and decline of trees (Derrick and Barthe, 2000). Trees propagated from psorosis B-infected buds are severely stunted and may die within 3 to 6 years (Milne et al., 2003).

Symptoms

The classical symptom of psorosis in adult citrus trees in the field is bark-scaling in the trunk and main branches (Fig. 2) and internal wood staining (Fig. 3) in the trunk and limbs (Roistacher, 1993). Gum may accumulate underneath the scales and impregnate the xylem, producing wood staining and vessel occlusion. Two types of psorosis have been described. The more common psorosis, form A, bark scaling first appears on trees 10 to 15 years old and is restricted to the main trunk and limbs. Old leaves are usually symptomless, but chlorotic flecks may occur in young leaves. With the more aggressive form B of psorosis, there is extensive bark-scaling that affects thin branches, symptoms appear earlier, such as chlorotic blotches, blisters (Fig. 4) or gummy pustules on the underside on old leaves, and, sometimes, there are irregularly depressed ringspots on the fruit rind (Fawcett and Bitancourt, 1943; Roistacher, 1993).

Figure 2. Psorosis symptoms of bark scaling on the trunk of sweet orange. Photo courtesy of C. N. Roistacher.

Known Hosts

Psorosis is mainly a disease of sweet orange, mandarin and grapefruit, with sweet orange being the most susceptible. CPsV has been serologically detected in varieties of sweet orange (*Citrus sinensis*), sour orange (*C. aurantium*), lemon (*C. limon*), grapefruit (*C. paradisi*), clementine (*C. clementina*), satsuma (*C. unshiu*) and some mandarin hybrids, like Fortune mandarin (*C. clementina x C. tangerina*) and Ortanique tangor (*C. reticulate x C. sinensis)*; psorosis will probably infects other species as well (Milne et al., 2003). Some of these

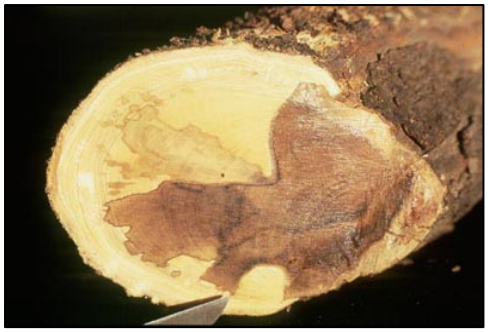


Figure 3. Staining (diagnostic symptom) of sweet orange trunk infected with psorosis form A. Photo Courtesy of C. N. Roistacher

Figure 4. Psorosis B induced blister lesions on the lower side of a sweet orange leaf. Photo Courtesy of EcoPort. (http://www.ecoport.org)

species, like sour orange, lemon or rough lemon, do not develop bark scaling, although they may show intense young leaf symptoms. *Poncirus trifoliate* can harbour CPsV symptomlessly (Milne et al., 2003).

Known Distribution

Citrus psorosis has been confirmed in North and South America, South Africa and the Mediterranean basin (Milne et al. 2003; Roistacher, 1993). Psorosis-like symptoms occur in most citrus-growing regions worldwide; however, the development of reliable detection methods for CPsV is recent. Currently, many countries have not confirmed CPsV by biological, serological or molecular indexing (Milne et al., 2003).

Distribution within the US

Psorosis has been the most serious virus disease in the Lower Rio Grande Valley, Texas. The disease has been an occasional problem in old citrus plantings in Arizona (Olsen e. al., 2000). Psorosis is known to be present in old Californian citrus trees. The use of nucellar virus-free and certified budwood free of psorosis has practically eliminated the disease (Derrick and Barthe, 2000). Some older trees are still around with psorosis, but it is quite rare in more recent plantings (Derrik, 2005, personal communication).

Survey

Currently, there are no specific survey methods for this disease. Symptoms are not specific enough to be used alone for surveying purposes. Apart from CPsV, other diseases and disorders can cause bark-scaling resembling those of psorosis, such as Bahia bark scaling, leprosis, foot rot caused by *Phytophthora*, Rio Grande gummosis, psorosis-like bark scaling, and genetic disorders, such as lemon shell bark and sunscald; thus, diagnosis of psorosis by bark scaling alone is not reliable.
Psorosis is currently diagnosed by biological indexing, serology, and molecular techniques. Biological indexing for psorosis is done by grafting tissue to young seedlings of sweet orange and keeping them in a cool greenhouse (18 to 24 °C). Symptoms do not develop at higher temperatures. CPsV causes a shock reaction with leaf drop and shoot necrosis in the first flush (Fig. 5), and a chlorotic leaf flecking and spotting in successive flushes. A cross-protection test, using psorosis B as a challenge, is required for a more specific diagnosis. First, sweet orange seedlings are graft-inoculated with tissue of the candidate tree, and are later grafted with a source of psorosis B. Psorosis-free plants develop the severe psorosis B symptoms after 6 months, whereas in psorosis A-infected plants such symptoms fail to develop (Roistacher, 1993; Milne et al., 2003).

Some isolates of CPsV can be mechanically transmitted to diagnostic herbaceous species (Derrick et al., 1988; da Graça et al., 1991). Sweet orange seedlings (cv. Pineapple or Madame Vinous) show shock and/or chlorotic flecks in young leaves. *Chenopodium quinoa* initially reacts with chlorosis and necrotic local lesions in 4 to 10 days. *Gomphrena globosa* reacts with necrotic local lesions with red halos in 10 days, followed by systemic necrosis (Milne et al., 2003).

Among the serological methods, Djelouah et al. (2000) developed antiserum and monoclonal antibodies that are highly effective in detecting numerous strains of the psorosis virus from many sources and countries. Alioto et al. (1999) improved the detection of the psorosis virus by using monoclonal and polyclonal antibodies. The direct tissue blotting immunoassay (Martin et al., 2002) and the immunosorbent electron microscopy have also been successfully used (Martin et al., 2004). The retrotranscription, polymerase chain reaction (RT-PCR), and molecular hybridization have also been used (Martin et al., 2004). There is a remarkable correlation between the detection of psorosis by biological indexing, ELISA, molecular hybridization, RT-PCR and immunosorbent electron microscopy (D'Ongia, 1998; Martin et al., 2004; Roistacher et al., 2000).

Figure 5. Shock symptoms caused by psorosis A inoculated to sweet orange grown at 24 to 27ºC (left) and at 32 to 38ºC (right). No symptoms develop at warmer temperature. Photo Courtesy of EcoPort (http://www.ecoport.org)

Key Diagnostics

Symptoms include the presence of the classical bark-scaling, internal wood staining, blisters on leaves, stems and fruit. Other methods include shock reaction on indicator plants, cross protection with psorosis B, protein molecular mass of 48 kDa, reaction to ELISA antiserum, and RT-PCR reaction.

Citrus tristeza closterovirus

Scientific Name
Citrus tristeza closterovirus

Common Name
Tristeza, seedling yellows, quick decline, CTV

Type of Disease
Plant pathogenic virus

Reason for inclusion in manual
National pest list and Emerging plant pest list

Pest Description
CTV is a closterovirus with long flexuous particles (2000 x 11 nm in size) containing a non-segmented, positive-sense, single stranded RNA genome. It is a phloem-limited virus.

Pest Importance
CTV is the most destructive viral disease of citrus and is not easily diagnosed. It is the most economically important pathogen of citrus worldwide. It has caused the death of infected trees of most citrus cultivars (except lemons) grafted on the highly susceptible sour orange rootstock. Millions of trees had been destroyed by the disease in the US (> three million), Argentina (ten million), Brazil (> six million), and other countries; thus, accurate survey data on the distribution and incidence of CTV-infected trees are important for control efforts.

Symptoms
Symptoms are affected by the CTV isolate and environmental conditions. Most CTV isolates cause stunting, leaf cupping, vein clearing and chlorosis. Trees with a severe strain may quickly decline and die. Severe stem pitting or honeycomb in limes, grapefruit, and sweet orange are often seen at the bark patch below the bud union. Fruits from infected trees are usually small and of poor quality. Characteristic symptoms are shown in Fig. 1.

Figure 1. Symptoms of citrus tristeza virus. Photo courtesy of T. Gottwald.

Transmission
CTV is transmitted by graft. In nature it is semipersistent by aphids, such as the melon aphis, *Aphis gossypii*; the green citrus aphid, *Aphis spiraecola*; and the newly introduced brown citrus aphid, *Toxoptera citricida*. The brown citrus aphid is the most efficient vector.

Known Hosts
Most species of citrus, and some species in other genera of the family Rutaceae (*Aegle marmelos, Aeglopsis chevalieri*, *Afraegle paniculata*, *Citropsis guilletiana*, *Fortunella margarita*, *Microcitrus australis*, and *Pamburus missionis*), serve as hosts for CTV. Trifoliate orange clones and many of their hybrids appear are resistant to infection.

Known Distribution
CTV is widespread throughout the tropical citrus-growing area. It has been found in all South and Central American countries. In the US, it is widespread in southern California and Florida, and restricted in Arizona, California, Hawai, Florida, Louisiana, and Texas.

Survey
CTV causes problems in nurseries and commercial groves. The optimum temperatures for virus infection and multiplication are 20 to 25ºC. Severe CTV epidemic may be expected by the presence of *Toxoptera citricida*, the brown citrus aphid.

Several methods are available for the survey of systemic infections by CTV. A single survey may examine all trees in an orchard. The “hierarchical sampling” developed by Hughes and Gottwald (1998, 1999) appears to be most widely used. In the hierarchical sampling, a block of citrus trees is divided into groups of four; each group is arranged in a two by two rectangle of four trees. Five young shoots (from the last flush), or fruit peduncles, or 10 expanded leaves, or 5 flowers (OEPP/EPPO, 2004), are collected around the canopy of each tree within a group and bulked for the determination of CTV. To start sampling, one out of the first four groups is selected at random, at the beginning of the block (grove); then, every fourth group of four is systematically sampled. It is important, to record the location of the groups, to return later if a group is found to be positive for CTV. Hierarchical sampling yields CTV incidence at the group level, but incidence at the scale of individual trees is calculated by means of a formula (Hughes and Gottwald, 1998, 1999). Surveys may be conducted when titer of CTV is known to be high. It was found that in California, titer of the CTV dropped in August and September to levels non-detectable by ELISA test.

Surveys in nurseries that supply propagation materials are important in preventing the spread of the disease locally or over long distances. Standard sampling for nursery plants include the collection of two young shoots or four leaves.

Key Diagnostics
The classic identification procedure for CTV is to graft-inoculate seedlings of Mexican lime (C. *aurantifolia*) and observe the development of vein clearing, leaf cupping and stem pitting. The development of DAS-ELISA (Garnsey and Cambra, 1991), tissue print-ELISA (Garnsey et al., 1993; Cambra et al., 2000), and other molecular techniques has allowed the test of thousands of samples for CTV infection.

Nematodes

Meloidogyne spp.

Scientific Name
Meloidogyne citri
M. donghaiensis
M. fujianensis
M. indica
M. jianyangensis
M. kongi
M. mingnanica

Common Name(s)
Asian citrus root-knot nematodes

Type of Pest
Nematode

Taxonomic Position
Phylum: Nematoda, **Order:** Tylenchida, **Family:** Meloidogynidae

Reason for inclusion in manual
National pest list

Pest Description
In Asia, a complex of root-knot nematodes, referred to throughout this document as Asian citrus root-knot nematodes, attacks the roots of *Citrus* crops. We have elected to address the entire complex of nematodes in a single document as information on each species is scarce. The Asian root-knot nematodes addressed in this document are not known to occur in the U.S. These nematodes should not be confused with *Meloidogyne incognita*, the “citrus root-knot nematode” that occurs in the U.S. or *Tylenchulus semipenentrans*, the “citrus root nematode” that is an important pest of *Citrus* spp. in the southern U.S. (Davis and Venette, 2004).

Figure 1. Example of a perineal pattern of *M. incognita* (common in US). Photo courtesy of H. Ferris.

The perineal pattern (Fig. 1), a unique and complex structure located at the female posterior body region, is comprised of the vulva-anus area, tail terminus, phasmids, lateral lines and surrounding cuticular striae; it is the primary distinguishing feature of these plant parasitic nematodes. Other distinguishing characteristics are listed in Table 1 (Inserra et al., 2003a-g; Davis and Venette, 2004).

Table 1: Characterisitcs of seven species of Asian root-knot nematodes

SPECIES	PERINEAL PATTERN	STRIAE	DORSAL ARCH	LATERAL LINES	OTHER INFORMATION
M. citri	oval	coarse and fine striae	low or moderately high	indistinct	
M. donghaiensis	rounded	coarse and fine striae	low or moderately high	indistinct	
M. fujianensis	oval	coarse and fine striae	moderately high	indistinct	A small swelling with a central pit is present outside the edge of the vulva.
M. indica	faint pattern	faint striae forming a distinct whorl in the tail area extending between the vulva and	low	indistinct	Second stage juveniles (J2) have tail terminus tapered with a broad and rounded tip.
M. jiangyanensis	rounded	coarse and fine striae	low or moderately high	indistinct	
M. kongi	ovoidal	fine stria	low or moderately high	indistinct	
M. minganica	ovoidal with a very distinct inner area, which contains vulva and anus	few coarse striae in an eight-shaped figure with a large base and a small top marked fine striae.	low or moderately high	indistinct	

All seven species of Asian citrus root-knot nematodes have sedentary endoparasitic habits. Second stage juveniles (J2s) penetrate host roots in the soil, where they establish a specialized feeding site (giant cells) in the stele. As the J2s develop, they cause the roots to swell, and females will become swollen. Females rupture the root cortex and often protrude from the root surface with egg masses. J2s emerge from the egg masses and migrate in the soil (Inserra et al., 2003a-g).

The nematodes are dispersed through root material, soil debris, and poorly sanitized bare root propagative material.

Biology and Ecology (from Davis and Venette, 2004)
Collectively, little is known about the biology of Asian citrus root-knot nematodes. Presently, *Meloidogyne fujianensis* is best described. Pan et al. (1999) have investigated

the phenology and biology of *M. fujianensis* on *Citrus reticulata* in Nanjing County (Fujian Province), China. *Meloidogyne fujianensis* is active year-round in this region;peak infection occurs between September and October and then again in the following season between March and April (Pan et al. 1999). During these periods, juveniles of various stages may be found in the soil. The life span of *M. fujianensis* lasts 55 to 60 days at 25°C, with 30 to 35 days spent from root infection by second stage juveniles to egg production by mature females. In a separate, differential host study by Vovlas and Inserra (2000), *M. citri* egg masses were initially found on satsuma, sour orange and tomato, 28 to 35 days following root penetration.

Development time varies depending on temperature, host availability and other biotic and abiotic factors. Population density appears to be correlated to the increasing air temperature; peaks have been observed during periods of active *Citrus* root growth in the spring and fall (Pan et al. 1999).

Adult: According to Pan et al. (1999), two months after a reported minimum of 308.4 degree days (°C), a population density peak of 117.6 females/2g roots occurred, compared to 94.3 females/2g roots observed in December, two months following a maximum of 891.3 degree days. Females swell, producing large gelatinous egg masses or sacs that contain very few eggs compared to other *Meloidogyne*. In December and January, during peak oviposition, 87.2 egg-sacs/2g roots and 150.2 egg-sacs/2 g of roots were reported, respectively. Only 5 of the 10 egg-sacs examined contained eggs; relatively few eggs were observed (1.2 eggs/female), considering the size of the egg sac matrix is 3 to 5 times the size of the female (Pan et al. 1999). The egg mass also serves as a protective padding. The egg sac is deposited on either galled root surfaces or inside root galls. *Meloidogyne fujianensis* primarily reproduces asexually, though some sexual reproduction must occur because males are present in the population. More males are observed during periods of drought stress, while more females are observed during more favorable conditions when the food supply is abundant (Pan et al. 1999).

Egg: Egg hatch may or may not involve stimulation from the host root. Hatching can occur for an extended period, depending on temperature. *Meloidogyne* eggs will not hatch under extended dry periods and may persist in soil or dry roots awaiting more favorable (moist) soil conditions.

Larva: For *Meloidogyne,* emergence occurs under moist soil conditions; juveniles may become inactive under dry conditions. *Meloidogyne* larvae can be easily distributed by irrigation ditches; they can survive in water saturated soil where larvae may survive under water for up to three weeks. There are four juvenile stages. The first stage occurs inside the egg. Following a molt and emergence, second stage juveniles move out of the egg and invade the host plant roots. The second stage is the only period when juveniles are mobile and are thought to be attracted to host plant roots.They may feed singly or in a group. If, after egg hatch, a larva cannot find a suitable feeding site on a host, it will continue searching until its energy is depleted. When a suitable site is selected, the larva will pentrate the root, usually near or behind the root cap, at lateral root initials or in galled root tissue near an embedded adult female. The site where one juvenile enters the root may attract others. The juvenile moves through the root to the region of cell differentiation, settles, and becomes inactive while feeding. Feeding induces cellular changes in the primary phloem or parenchyma, changing them into large, nutrient-rich cells from which

juveniles feed until development is completed. If large, specialized cell (gall) formation does not happen as a result of host infection, the larva may not complete its development and leave in search of another root or die of starvation in the process. When giant cell formation occurs, tissues surrounding the feeding nematode begin transforming at approximately the same time, producing a gall within 1 to 2 days following root penetration. The larva will swell as it feeds until development is complete. The total development time varies depending on temperature.

Pest Importance

Because *Meloidogyne* spp. are generally considered to be the most economically damaging nematodes (and not just *Citrus* to spp.), these nematodes may have the potential to severely affect citrus production if the Asian root-knot nematodes arrived and established in the U.S. In a series of assessments, *M. citri, M. donghaiensis*, *M. fujianensis*, *M. indica*, *M. jiangyanensis*, *M. kongi*, and *M. mingnanica* received medium risk ratings (Inserra et al., 2003 a-g). The high value of citrus elevated the ratings, but limited information on the potential degree of economic damage and the basic biology of the nematodes tempered the assessments (Davis and Venette, 2004).

The economic impacts of a single species are difficult to measure because nematodes often occur in mixed populations (Davis and Venette, 2004); however, these nematodes are thought to pose a real economic threat to citrus producing regions, especially China and India, where estimated crop losses have been as high as 20 to 50% (Pan, 1985). *Meloidogyne fujianensis*, widespread in the Fuijan region of China after which it is named, reportedly causes more damage than the better known citrus nematode, *Tylenchulus semipenetrans* (Pan et al. 1999).

Symptoms

Damage to host plants caused by root-knot nematodes includes impaired root growth (e.g., small gall formation) (Fig. 1), proliferation of lateral roots, the stimulation of giant cell growths at feeding sites in parenchyma and phloem, and impaired root function (contributing to chlorosis, stunted growth, nutrient deficiencies, and/or necrosis of above-ground plant parts). Symptoms of nematode damage may be similar to those caused by nutrient or water deficiency. Nematode infestation of plant roots limit water uptake. Infested plants may appear wilted under hot and sunny conditions, even with ample soil moisture. Symptoms may not be apparent until plants reach later stages of growth. Injured root tissue is susceptible to other disease-causing pathogens. Much of the visible damage to plant hosts is likely caused by a combination of biotic and abiotic factors.

Specific damage to *Citrus* caused by *Meloidogyne* spp. [documented for *M. fujianensis*] includes the yellowing of leaves, premature flower drop, early ripening and fruit drop, and reduced fruit quality and yield. Citrus roots infected by root-knot nematodes show swollen rot tips and axes. Nematode egg masses are often visible on the surface of galls.

Figure 1. Galled roots caused by Asian citrus root-knot nematodes. Photos courtesy of R. Inserra.

Known Hosts

Meloidogyne citri infects and reproduces on citrus, such as Satsuma (*Citrus unshiu*), sour orange (*C. aurantium*), mandarin orange (*C. reticulata*) and trifoliate oranges (*Poncirus trifoliate*). The only known non-citrus host is tomato (*Lycopersicon esculentum*). *Meloidogyne donghaiensis, M. fujianensis, and M. jiangyangensis* infect and reproduce on citrus, such as mandarine orange. The only known non-citrus host for *M. fujianensis* is cogongrass (*Imperata cylindrica*). *Meloidogyne indica* infects and reproduces on lime (Citrus aurantifolia), sweet orange (*C. sinensis*), and morinda (*Morinda officianalis*). *Meloidogyne kongi* infects and reproduces on *Citrus* spp. and experimentally on pepper (*Capsicum spp.)*. *Meloidogyne mingnanica* infects and reproduces on Satsuma, trifoliate orange, and experimentally on sour orange

Known Distribution

These plant parasitic nematodes are only known to regionally occur within China and India, in areas where the climate ranges from tropical-subtropical to temperate. All seven species of Asian citrus root knot nematodes are known to be distributed in China. *Meloidogyne citri* has been reported from Shuinan, Shunchang, and Xiasha orchard locations in the Fujian Province. *Meloidogyne donghaiensis* has only been reported from the Fujian Province. *Meloidogyne fujianensis* has been repored from Nanjing County of the Fujian Province. *Meloidogyne indic*a has been reported from Wuping County of the Fujian Province. *Meloidogyne jianyangensis* has been reported from Jianyang County of the Sichuan Province. *Meloidogyne kongi* has been found in the Guangxi Province. *Meloidogyne mingnanica* has also been reported from the Fujian Province. *Meloidogyne indica* is also known to be present in Delhi India.

Potential Distribution within the U.S.

Asian citrus root-knot nematodes have a narrow host range, feeding primarily on Citrus hosts. Not all members of the Asian citrus root knot nematode complex are equally likely to find a suitable climate in the continental U.S. (Table 2). *Meloidogyne citri*, *M. donghaiensis*, *M. fujianensis*, *M. indica*, and *M. mingnanica* are tropical species that are likely to find a suitable climate only in southern Florida. In comparison, *M. jianyangensis* and *M. kongi* are more temperate species that are likely to be found in the climatically suitable regions of the U.S.

Table 2: Habitat zones for Asian citrus Root-Knot Nematodes (Taken from Davis and Vennette, 2004).

Nematode(s)	Biomes/Habitat Zones	Estimated % of the continental U.S. that could provide suitable climate for establishment
Meloidogyne citri *M. dongahaiensis* *M. fujianensis* *M. mingnanica*	Tropical and subtropical broadleaf forests[1]	<1%
M. indica	Tropical and subtropical broadleaf forests[1] Tropical and subtropical dry broadleaf forests[2]	<1%
M. jianyangensis	Temperate coniferous forests	19%
M. kongi	Temperate broadleaf and mixed forests Tropical and subtropical broadleaf forests[1]	28 to 29%

[1] Collier, Dade, and Monroe Counties in southern Florida have this climate zone.
[2] This biome does not occur in the U.S.

Survey

It is known that infection and development of root-knot nematode species are favored by coarse-textured soils, which are low in organic matter. Survey efforts should be focused in soils of this type. Current techniques for nematode sampling should prove adequate to detect most infestations of new *Meloidogyne* spp. The success of methods depends heavily on the amount of sampling that can be conducted. If only a modest sampling effort can be made, the likelihood of detecting infrequent, sparse infestations of these nematodes is low.

Vovlas and Inserra (1996) outline general considerations for conducting a survey for new *Meloidogyne* spp. In general, they recommend sampling root tissues to inspect for the presence of galled roots (Fig. 1). During routine surveys for nematodes in citrus orchards, abnormally swollen fibrous roots should be placed in plastic bags with soil from the rhizosphere and submitted to a nematology diagnostic lab for nematological analysis. Microscopic examination of the roots is necessary to separate galls induced by root-knot nematodes from tip swellings caused by sting and dagger nematode feeding. The occurrence of root-knot nematodes on weeds is common in citrus groves. Soil samples from these groves are positive for root-knot nematodes because they originate from weed hosts. Careful examination and separation of roots in the samples provide certainty of the origin of nematode infestation. Root-knot nematode surveys based only on nematological analysis of soil provide only an indication of nematode presence, but not of hosts.

From Davis and Venette (2004): Alternatively, soil samples may be collected. General principles described by Greco et al. (2002) apply to *Meloidogyne* spp. Samples of soil or

host roots must be collected with the purpose of obtaining males, juveniles, or nematodes within root tissues. Samples must then be processed to separate nematodes from soil and debris. Finally, nematodes must be prepared either for identification using morphological (e.g., perineal patterns) or molecular techniques. Soil sampling is typically based on the collection of cylindrical cores of soil. Frequently, a sample unit is composed of several cores that are thoroughly combined and mixed. The number of sample units collected from a field is the sample size. Not all soil from each sample unit will necessarily be processed; rather, nematodes will frequently be extracted from a soil subsample.

Sampling may be conducted to detect the presence of new *Meloidogyne* spp. in an individual field or over a broader geographic area. For quarantine nematodes that are known to occur in the U.S. (e.g., *Globodera rostochiensis*), it may be important to take sufficient samples to certify with a high degree of confidence that the probability of a nematode species being present in an individual field is very low. To achieve this goal, highly intensive sampling may be needed. Been and Schomaker (2000) proposed a sample unit of 50 cores (presumed to be 1 in diameter x 6 in deep) collected on a 5 m x 6 m (~16 ft x 20 ft) grid. This sampling procedure results in the collection of 2 kg soil per sample unit; a sample size of 6 to 7 units per hectare is recommended. Such a high level of sampling intensity provides a ≥90% probability of detecting nematode aggregations with ≥200 cysts/kg soil at their center. The sampling recommendations of Been and Schomaker (2000) are based on empirical observations of the size of nematode patches (or foci) when they occur in potato fields; nevertheless, the same principles should apply to surveys for *Meloidogyne* spp. The protocol should have a high probability of detecting members of the genus when they are present in a field.

In contrast, it may be more valuable (and perhaps even more cost effective) to use a smaller sample unit and/or sample size per field to maintain a high probability of finding an exotic nematode somewhere within a geographic area, even though the likelihood of finding a species in an individual field might be lower. For regional surveys of nematodes, Prot and Ferris (1992) recommend a single composite sample of 10 cores per field. Cores should be collected approximately 55 m (180 ft) apart throughout the entire field. For most field and forage crops, soil samples should be collected at a depth of 15 to 40 cm (6 to 16 inches) within the root zone. Samples should be collected with an Oakfield- or Veihmeyer sampling tube (~1 inch inner diameter). Soil samples should be collected from fields that include one or more hosts in the cropping rotation. The sampling recommendations from Prot and Ferris (1992) were based on observations from cotton and alfalfa. The sampling protocols have not been evaluated orchards, but the principles upon which the recommendations are based should still apply. A 10-core, composite sample is particularly efficient at detecting nematodes when species are “frequent and abundant.”

The number of fields that should be sampled to maintain a high probability of detection within a region depends on the chances that nematodes are found in an individual field. The chances that a nematode species will be detected when it is present within a field are influenced a number of factors; soil type; vertical distribution of nematodes within the soil profile; time of year; the number of soil samples that are collected; the unit size of those samples; the amount of soil that is processed (typically a subsample of the sample unit); and the method(s) of nematode extraction and identification. The vertical distribution of

new *Meloidogyne* spp. is likely to be influenced by the distribution of roots. Sub-sampling and extraction efficiency also affect the likelihood of detecting a nematode when it is present in a sample. Both factors reduce the likelihood that nematodes will be detected when they are present.

Root knot nematodes are extracted from soil using a variety of techniques. Six methods (and subtle variations thereof) are particularly common: Baermann trays; Baermann trays with elutriation or sieving; centrifugal flotation; flotation-sieving; semiautomatic elutriation; and Cobb's decanting and sieving. These methods are described in detail by Barker (1985) and will not be repeated here. The efficiency of nematode extraction is influenced by the amount of soil that is processed at one time. Extraction efficiencies are greatest when 100 g (~70 cc) to 450 g (~300 cc) of soil are processed. Extraction efficiencies for *Meloidogyne* spp. are frequently low and can vary between 13 and 45%.

Parasitic Plants/Weeds

Cissus verticillata

Scientific Name:
Cissus verticillata Nicolson & C.E.Jarvis

Synonyms:
Cissus sicyoides

Common Name:
Seasonvine, possum grape, waterwhite treebine

Type of Pest:
Plant

Taxonomic position:
Phylum: Anthophyta, **Order:** Rhamnales, **Family:** Vitaceae

Reason for inclusion in manual
Texas PPQ indicated that this weed was becoming a problem in citrus groves in the Lower Rio Grande Valley

Pest description:
Possum grape is an elongated, perennial vine. All parts of the plant are odorless unlike the malodorous *C. incisa,* which is common in Texas. Immature stems are dark green and glabrous. Mature stems have swollen nodes and are characterized by a thin, peeling bark that is covered with reddish papillae and cream-colored lenticels. The peeling bark exposes dark green stems.

The leaves are typically large, simple, alternate, glabrous, and succulent (Fig. 1A). They range from broadly rounded to ovate with rounded or cordate leaf bases. The margins are entire and the petioles are elongated. Conspicuous, firm tendrils are opposite some leaves and are present at the shoot apex. In contrast, *C. incisa* has much smaller leaves, and the margins are irregularly toothed.

The inflorescence is a densely flowered cyme that extends from the leaf axils. The calyx is light green, cup-shaped, and forms a rim around the ovary. The corolla includes four, yellow-green, connate petals that are attached to a floral disc. The androecium consists of four distinct white stamens. The pistil has one unbranched style and is subtended by a nectariferous disc. The superior ovary develops into a succulent black or purple berry that is similar to a small grape (Fig. 1B).

The fleshy, grape-like fruits have been distributed by migratory birds. The combination of bird dispersal and the adjacent canal system may prove to be important vectors to disperse this aggressive species to other citrus production centers in the Lower Rio Grande Valley of Texas

Figure 1. Photographs of *Cissus verticillata* in four settings: close-up of the leaves (A), the mature black to purple berries (B), engulfing a large live oak tree (C), and on an orange tree (D). Photos courtesy of J. H. Everitt, USDA, ARS.

Economic impact

Little information is given concerning the economic impact of this vine.

This perennial vine is native to tropical Mexico, Central America, and the Caribbean; it has recently been rediscovered in the Lower Rio Grande Valley of Texas. A dense population of this exotic species has been located in a brushy area along a canal network and in two adjacent citrus groves near Weslaco. This species produces a dense mantle that covers other vegetation, appears to be invasive, and may pose a potential weed problem in citrus in the Lower Rio Grande Valley.

Known hosts

Quercus virginiana (live oak), *Salix nigra* (black willow), *S. exigua* (sandbar willow), *Melia azedarach* (Chinaberry), *Sapium sebiferum* (Chinese tallow), and *Carica papaya* (papaya).

Known distribution

Standley (1923) listed seven species of *Cissus* (Vitaceae) in Mexico including *Cissus verticillata,* which is distributed throughout most of the tropical regions of Mexico, Central America, and the Caribbean (Standley, 1923). Only one species, *C. incisa* Des Moulins, is common in Texas. *Cissus verticillata* was initially reported in Texas and Florida by Vines (1960); however, it was not listed by Correll and Johnston (1970), Hatch et al. (1990), Jones et al. (1997) or Jones and Wipff (2003). Although this species is not widespread in subtropical Texas, there probably are no barriers to its spread.

Potential distribution within the USA

Lower Rio Grande valley, Texas, the citrus production region of Florida

Survey

A dense population of *C. verticillata* was found in anthesis and fruiting conditions in Hidalgo County, Texas, 4 November 2003, in a field reconnaissance along a canal network, in addition to two citrus groves near Weslaco; this represents the first record of this species on citrus. The vine produces a dense, "kudzulike" mantle that covers *Quercus virginiana* (live oak) (Fig. 1C), *Salix nigra* (black willow), *S. exigua* (sandbar willow), *Melia azedarach* (Chinaberry), *Sapium sebiferum* (Chinese tallow), and *Carica papaya* (papaya). Propagule dispersal has allowed *C. sicyoides* to encroach onto a grapefruit and orange grove south of the canal (Fig. 1D). In its early growth stages, the vine is difficult to distinguish from the dark green foliage of the citrus canopy. Once established, vines completely shrouded many of the trees; as a result, the citrus grower was forced to launch a vigorous vine removal program in both groves.

Cuscuta reflexa

Scientific Name:
Cuscuta reflexa Roxb.

Synonyms:
Cuscuta elatior, *C. gigantean, C. grandiflora, C. hookeri, C. macrantha*, *C. macrantha*, *C. megalantha*, *C. megalantha*, *C. reflexa var. grandiflora*, *C. verrucosa, Monogynella reflexa.*

Common Name:
Giant dodder

Type of Pest:
Parasitic plant

Taxonomic position:
Phylum: Magnoliophyta, **Order:** Solanales, **Family:** Cuscutaceae.

Reason for inclusion in manual
National Pest list and Regulated plant pest list

Pest description:
Giant dodder is an annual stem parasite with leafless, thread-like, orange, red, or yellow stems that twine over other plants (Fig. 1). Dodder can be problematic in agricultural crops, especially alfalfa and tomatoes. In addition, dodder seed is difficult to exclude from commercial alfalfa, clover, or flax seed.

The plant is vigorous, typically attacking woody plants. Known infestations have been eradicated in California. Giant dodder is native to Asia, where it is a serious pest in citrus, coffee, peach, forest trees, and many others.

Figure 1: Giant Dodder. Photo by Gerfried Deutsch.

Seedlings: Lack cotyledons. Develop a small temporary root to support a thread-like shoot, 4 to 10 cm long. Shoot moves slowly in a circular pattern as it grows until it touches a support. Upon contact with a suitable host, knob-shaped organs (haustoria) develop to penetrate the host stem. Seedlings die without a host within 10 days to several weeks depending on the species.

Mature plant: Stems 1 to 2 mm thick, glabrous, lack leaves or have appressed, scale-like leaves about 2 mm long; red, yellow, or orange, but contain some chlorophyll and are sometimes tinged green. Growing stems branch and attach to new host stems with haustoria. Each dodder branch obtains nutrients from the host independent of older branches. One plant can cover 10 to 15 ft^2. Roots and underground structures are modified into specialized knob-shaped organs (haustoria) that penetrate host stems.

Figure 2. Giant Dodder in flower. Photo by Gerfried Deutsch

Flowers: Time variable depending on species, but generally May to October. Bell-shaped. Corolla white with purplish rim, 6 to 10 mm long. Calyx ~ ¼ corolla length. Style 1, very short, with 2 linear stigmas longer than the style (Fig. 2).

Fruits and seeds: Capsules are spherical, ovoid, or conical, opening irregularly or like a lid at the top (circumscissile). Seeds 1 to 4, spherical, oblong, ovoid, or angular. Capsules conical, 5 to 8 mm long. Seed 3 to 3.5 mm long. Postsenescence characteristics: Frost kills plants, but haustorial tissue of some species can overwinter in host stems and develop new stems the following spring. Stems do not persist through winter.

Mostly found in natural communities, although they sometimes infest nursery crops, landscaped sites, and agricultural crops, especially alfalfa, clovers, and tomatoes. The host range is broad, but monocots (excluding asparagus) are seldom affected.

Reproduces by seed and vegetatively. Broken stems can develop new haustoria. Seeds can disperse by water, animal ingestion and movement, and human activities and machinery. A proportion of seed has a hard coat that must be weakened by scarification, microbial decomposition, and winter chilling before germination can occur. Germination does not require the presence of a host plant. Under favorable conditions, seed can germinate in the fruits. Seed can remain viable for at least 10 years in the soil. Emergence is typically from the top 5 cm of soil. In most years, the period of emergence ends by mid-May in the Central Valley.

Pest Importance

Cuscuta reflexa is less widespread than *C. campestris*, but is capable of serious crop damage where it does occur. It is classified as a 'principal' or 'serious' weed in Afghanistan, Nepal, India and Pakistan. *Cuscuta reflexa* is distinguished from *Cuscuta campestris* and other species by having only a single style. Other weedy species with a single style can be ruled out when the style is shorter than the stigmas. *Cuscuta monogyna, C. lupuliformis and C. japonica* have styles longer than the stigmas, and flowers less than 5 mm long.

Symptoms

Giant Asian dodder is a yellow-green vine that resembles spaghetti (Fig. 3). It is able to parasitize a wide range of hosts including many agricultural crops (alfalfa, tomatoes, and onions, among others). Giant Asian dodder has been observed parastizing 20 host plants in southern Texas ranging from herbaceous plants to woody ornamentals (live oak, crape myrtle and Ligustrum). Giant Asian dodder seeds are the size of coffee grains and have thick seed coats that are impermeable to oxygen and water; this allows the seed to remain viable until suitable conditions are present for germination. Dodder seed can remain viable in the soil for a period of 10 to 20 years. Usually most seed that is produced will germinate the following year.

Figure 3. This is an Arizona ash in south Houston that is being aggressively parasitized by giant Asian dodder. Photo courtesy of K. Camilli. http://www.texasurbantrees.org/news/asiandodder_html.php

Dodder seedlings have been reported to emerge from depths of 4 inches in the soil, although usually most seedlings originate from seeds located at a depth of ½ inch or less. Once the dodder seeds germinate, a rootless and leafless seedling is produced. The seedling absorbs water initially from the soil, in a manner similar to a root. The seedling uses the soil as an anchoring point to find a nearby host to parasitize. Giant Asian dodder seedlings can survive for a period of several weeks without a host plant. In this time period, the seedling extends in length and rotates counterclockwise, seeking a host for a source of nutrients and water. If the seedling does not find a host within a few weeks, it dies.

The second method of overland spread is by fragmentation. This occurs when sections of the dodder vine itself are removed from the main plant and become established on another host. Haustoria are produced from these fragments, establish a connection to the host to obtain necessary nutrients, and continue to grow.

Known hosts

Primary hosts include *Citrus medica* (citron), *Coffea arabica* (arabica coffee), *Litchi chinensis* (leechee), and *Prunus persica* (peach). It can also parasitize *Desmodium spp.* (Tickclover), *Rubus spp.* (Blackberry), and *Viburnum spp.* *Cuscuta reflexa* is known to kill *Vitex negundo* (Chinese chastetree) and Ziziphus jujube (Jujube).

Known distribution

Giant dodder is mainly confined to tropical Asia, but has been recored in Afghanistan, Bangladesh, Bhutan, China, India, Indonesia, Nepal, Pakistan, Sri Lanka, and Thailand. It

has only been reported in Texas in the United States. A previous infestation on the south coast of Los Angeles County has been reported.

Potential distribution within the USA

A major concern is the potential for this plant parasite to vegetatively spread through infected plant material and incoming cargo to local ports. The key word is ‘potential’, because so far the only known cases of Giant Asian Dodder are five locations in southeast Houston, Texas. Because this parasitic plant has such a broad host range, the possibility for further spread of this plant exists.

Survey

Growing at a rate of six inches per day, the giant Asian dodder can rapidly spread from one infected plant to another nearby host. It can spread to a new area by two other methods, seed production and fragmentation.

Giant Asian dodder is a yellow-green vine that resembles cooked spaghetti. When sampling from a site, the dodder is inspected to determine if it is Giant Asian Dodder. To determine if the plant is Giant Asian Dodder the color of the dodder is observed, the flowers, the thickness of the dodder and what host plants it is parasitizing. At each location, detailed information is recorded, including host plant species, health of the dodder, species, flowering, seeds produced, thickness of the dodder, etc. Once the information is collected, the dodder is sampled for a permanent press collection and identified by a trained plant taxonomist. Part of the host plant, as well as the dodder species itself, is pressed, along with flowers that are produced. The main way to identify the different dodder species is to dissect the flowers. They are very unique to each species and reliable for species identification.

Mollusks

Achatina fulica

Scientific Name
Achatina fulica Bowdich

Synonyms:
Lissachatina fulica

Common Names
Giant African land snail, African giant snail, Kalutara snail

Type of Pest
Mollusk

Taxonomic Position
Phylum: Gastropoda, **Order:** Stylommatophora, **Family:** Achatinidae

Reason for inclusion in manual
National Pest list and Eastern Region pest list

Pest Description
Achatina fulica is distinctive in appearance and is readily identified by its large size and relatively long, narrow, conical shell. Reaching a length of up to 20 cm, the shell is more commonly in the range of 5 to 10 cm. The color can be variable, but is most commonly light brown, with alternating brown and cream bands (Fig. 1) on young snails and the upper whorls of larger specimens. The coloration becomes lighter towards the tip of the shell, which is almost white. There are from six to nine spirally striate whorls with moderately impressed sutures. The shell aperture is ovate-lunate to round-lunate with a sharp, unreflected outer lip. The mantle is dark brown with rubbery skin. There are two pairs of tentacles on the head: a short, lower pair and a large upper pair with round eyes situated at the tip. The mouth has a horned

Figure 1. Adult Giant African Snail. Courtesy of M.A. Ciomperlik, USDA APHIS PPQ

mandible containing some 80,000 teeth. Eggs are spherical to ellipsoidal in shape (4.5 to 5.5 mm in diameter) and are yellow to cream in color.

Like most snails, *A. fulica* is hermaphroditic and, after a single mating, can produce a number of batches of fertile eggs over a period of months. Early reports of self-fertilization (van der Meer Mohr, 1949) have since been discounted. It lays eggs in batches of 100 to 400 with up to 1200 being laid in a year. These hatch after about 8 to 21 days under tropical conditions. Eggs are laid on the ground, often in the base of plants.

While the adult has an average lifespan of 5 to 6 years, it may live for as long as 9 years. It will readily enter a state of aestivation and can survive for years in this state. The tendency for a number of aestivating snails to be present in an area at any time, particularly when conditions are optimum for activity, can make control measures difficult. Although *A. fulica* is a tropical snail, it can survive cold conditions, even snow, by aestivating, though it is unable to establish itself in temperate regions. It is normally nocturnal and crepuscular in its habits, although it will become active in the daytime during rainy or overcast periods. This indicates that light, in addition to temperature, moisture and food, are all vital factors in snail activity.

Pest Importance

Achatina fulica is a polyphagous pest. Its preferred food is decayed vegetation and animal matter, lichens, algae and fungi. The potential of the snail as a pest became apparent after being introduced around the world into new environments (Rees, 1950). It has been recorded on a large number of plants, including most ornamentals, vegetables and leguminous cover crops, which extensively suffer. The bark of relatively large trees, such as citrus, papaya, rubber and cocoa, is subject to attack. Poaceous crops (sugarcane, maize, rice) suffer little or no damage from this species. There are reports of *A. fulica* feeding on hundreds of species of plants (Raut and Ghose, 1984). Thakur (1998) found that vegetables of the genus *Brassica* were the most preferred food item from a range of various food plants tested.

Symptoms

In garden plants and ornamentals of a number of varieties, and vegetables, all stages of development are eaten, leading to severe damage in those species that are most often attacked. Cuttings and seedlings are the preferred food items, even of plants such as *Artocarpus*, which are not attacked in the mature state. In these plants, damage is caused by complete consumption or removal of bark. Young snails, up to about 4 months in age, feed almost exclusively on young shoots and succulent leaves. The papaya appears to be the only fruit that is seriously damaged by *A. fulica*, largely as a result of its preference for fallen and decaying fruit.

Known Hosts

Achatina fulica is known to eat at least 500 different types of plants, including breadfruit, cassava, cocoa, papaya, peanut, rubber, and most varieties of beans, peas, cucumbers, and melons. Some of the primary hosts include *Arachis hypogaea* (groundnut), *Artocarpus* (breadfruit trees), *Brassica* (rape), *Carica papaya* (papaw), *Cucumis melo* (melon), *Cucurbita pepo* (ornamental gourd), *Dioscorea alata* (white yam), and *Musa* (banana).

Other known secondary hosts include *Daucus carota* (carrot), *Hevea brasiliensis* (rubber), and *Theobroma cacao* (cocoa).

Known Distribution

Bangladesh, Bunei, Cambodia, China (Hong Kong, Taiwan), Christmas Island, India, Indonesia, Japan, Malaysia, Maldives, Myanmar, Philippines, Singapore, Sri Lanka, Thailand, Vietnam, Africa (Côte d'Ivoire, East Africa, Kenya, Madagascar, Mauritius, Morocco, Réunion, Seychelles, Tanzania), U.S. (Hawaii), Barbados, Guadeloupe, Martinique, Saint Lucia, Brazil, American Samoa, Belau, Micronesia, Fiji, French Polynesia, Guam, Kiribati, Marshall Islands, New Caledonia, New Zealand, Marianas Islands, Papau New Guinea, Samoa, Solomon Islands, Tonga, Tulavu, and Vanuatu.

Potential Distribution within the US

In 1966, the snail was inadvertently released by individuals travelling from Hawaii to Florida, where it became established. Eradication efforts, which cost approximately $1 million, were successful. An anticipated distribution within the U.S. would include areas with subtropical to tropical climates.

Survey

Achatina fulica is a large and conspicuous crop pest that hides during the day. Surveys are best carried out at night using a flashlight, or in the morning or evenings following a rain event. It is easily observed, attacking plants that exhibit extensive rasping and defoliation. The amount and weight of *A. fulica* can break the stems of some host species. *Achatina fulica* can also be detected by signs of ribbon-like excrement and slime trails on plants and buildings.

Key Diagnostics

Achatina fulica is distinctive in appearance and readily identified by its large size and relatively long, narrow, conical shell. It can reach a length of up to 20 cm; the shell is commonly in the range of 5 to10 cm.

Theba pisana

Scientific Name
Theba pisana Mueller

Synonyms:
Helix pasana

Common Name:
White garden snail

Type of Pest:
Mollusk

Taxonomic Position
Class: Gastropoda, **Order:** Stylommatophora, **Family:** Helicidae

Reason for inclusion in manual
National Pest list and Eastern and Western Region pest lists

Pest Description
The shell is subglobose with a moderately depressed spire. The adult shell has 5 1/2 to 6 slightly convex whorls with shallow sutures. It is of medium size, ranging from 12 to15 mm (rarely to 25) in diameter, 9 to 12 mm (rarely to 20) in height, and is opaque and moderately solid. The umbilicus is narrow and partially to entirely covered by an expansion of the columella. The aperture of the shell is rounded and lunate and only slightly oblique. The lip of the aperture is sharp and unreflected, but some specimens show a thickening inside the lip. The juvenile shell has a sharp keel at the periphery, but in the adult shell the periphery is only slightly shouldered. The surface of the shell is not glossy, but is marked with many fine vertical striae. The background color of the shell is nearly always ivory white (rarely pink); there are often a variable number of narrow dark-brown spiral bands present. These bands may be solid, made up of dots and dashes, or absent. This difference in coloration does not have any systematic significance because it is apparently a polymorphic trait subject to differential selection pressures, correlating with microhabitat (Johnson, 1980). The first 1 1/2 whorls are generally dark in color, ranging from tan to dark brown, and give the appearance of a dot on the apex of the shell. This snail mates during the fall's rainy season in California. *Theba pisana* survives in a desert environment due to its protective covering over the shell case opening and estivates

Figure 1. White garden snail Theba pisana (Muller). Photo courtesy of P.M. Choate, University of Florida

during summer.

Biology and Ecology:
Garden snails are hermaphrodites, meaning that one individual possesses both male and female reproductive organs. Although they are able to self-fertilize, most snails mate with another snail. Reproduction takes place in early summer, and begins with pairing and courtship. After a period in which the members of the pair caress each other with their tentacles, each snail pierces the skin of its partner with a calcareous 'love dart', a spiny projection that is covered in mucus. The function of this love dart is unclear, but it is thought that the mucus may act to improve the survival of sperm. Mating then takes place, as each snail inserts its penis into its partner at the same time. The snails separate, and the sperm is stored internally until the eggs are ripe. After the eggs have been fertilized, the snails dig pits in the soil in which to lay the eggs. Hatchlings have translucent, delicate shells (2 mm) and will feed on tender shoots. Mature snails will feed on weathered wood and paper, using sand in their digestive tract. Snails live an average of 2 to 3 years, although some have survived four winters.

Pest Importance
This snail is particularly destructive to *Citrus spp.* It is a vector for the melon fungus, *Colletotrichum lagenarium*. The snail serves as a host for lungworm and other nematodes that are parasites of sheep and cattle. *Theba pisana* has a high reproductive capacity. It can survive long, arduous journeys on shipping containers because it can produce a wall of dried mucus over its opening, which reduces water losses during dormancy (aestivation). *Theba pisana* can cause severe damage and, occasionally, total destruction to legume based pastures (e.g., annual medics, lucerne, clovers) and seedling crops (e.g., wheat and barley). The re-establishment of pastures in snail infested areas is particularly difficult, as stock rejects pasture and hay that is heavily contaminated with snails (due to slime). This species is agriculturally important in southern Australia because it may climb onto the heads and stalks of cereals, beans, peas and grapes (for the raisin industry) in late spring/early summer to aestivate, or go into dormancy. When it attaches to cereals at harvest time, the crushed snails can block harvesters.

Symptoms
During dry weather, most slugs and snails aestivate hidden under logs, stones or buried in the earth. *Theba pisana* aestivates in the open on trees, fences, and other vertical surfaces. Pilsbry (1939) reported that *T. pisana* will mate following the early Californian rains in November. Like all helicid snails, *Theba pisana* is a cross-fertilizing hermaphrodite. The eggs are deposited several inches in the ground a few weeks after mating. Hatching occurs after a minimum of 20 days, but may occur later during dry weather. In the active season, this snail partially defoliates a variety of shrubs and trees, including citrus. The great density of the populations in California (up to 3000 snails per tree) and the rapid rate of reproduction are primary factors in making this snail a major pest (Mead, 1971).

Known hosts
Citrus spp., olive, and almond are primary hosts. Snails eat the foliage of many garden and ornamental plants and flowers. Vegetables include cabbage, carrots, cauliflower, celery, beans, beets, tomato, brussel sprouts, lettuce, onion, peas, radish, and turnips. *Theba*

pisana defoliates and chews on the tender bark of a variety of shrubs, including stone fruits, vines, olive trees, almonds and citrus, where they feed on twigs, fruit and blossoms.

Known distribution

Theba pisana is native to southwestern England and Wales, Ireland, western France, Switzerland, and the Mediterranean countries of Europe and Africa (Pilsbry, 1939; Burch, 1960). Its habitat in Europe is near the coasts (Kerney and Cameron, 1979). It has been introduced into the Atlantic islands, South Africa, Somaliland, and Western Australia.

Potential distribution within the USA

This snail was first noticed in North America in La Jolla, San Diego County, California, in 1914 (Chace, 1915; Basinger ,1923). It soon spread to several locations in Orange and Los Angeles Counties, but apparently was eradicated by 1940 (Hanna, 1966). Mead (1971) reported a second infestation in Los Angeles County in 1966; it was declared as eradicated in 1972. The snails were found and identified in August 1985 in San Diego, California at several localities in about a 10 square mile area. Hanna (1966) stated that *T. pisana* has been introduced into several eastern localities in the United States. There are no published records for North American populations outside of California. *Theba pisana* is also present in Bermuda, but has never been recorded in Florida or Hawaii.

Survey

A surveyor can follow slime trials. *Theba pisana* moves by means of a muscular foot; the mucus secreted by the foot aids with movement and leaves a tell-tale track. Snails show strong proclivity for climbing any vertical structure, preferably in the open when summer aestivation or dormancy starts. It aestivates, or passes summer, in a torpid condition in the open, or on trees, fences, plant stems, walls or any other vertical structure. *Theba pisana* will start mating during the rainy season. This snail is mainly nocturnal, but will emerge after rain during the day. They feed on a range of plant matter, and can be serious pests to gardens. This snail has a strong homing instinct, and spends the day, often in large groups, beneath stones and other structures when not aestivating. Densities of 3000 snails per tree have been recorded. They are easily transported on farm equipment due to their climbing nature. They hibernate through the winter in similar locations. Snails can be baited with metaldehyde.

Searches for *Theba pisana* should be directed to plants, fences, and other vertical surfaces in nature, especially in sandy areas. Crates shipped from areas where the snails are known to exist should be examined, especially lids and corners.

Glossary

Abdomen: The posterior of the 3 main body divisions of an insect; bears no functional legs in the adult stage.

Abiotic: Describes nonliving substances or environmental factors.

Acquisition feed: The feeding process by which an insect acquires a virus from an infected plant.

Acquisition time: The time required for an insect to acquire a virus from an infected plant.

Acrostichal: (Bristles) one or more longitudinal rows of small bristles along (in Diptera: flies) the middle of the dorsum of the thorax.

Aculeus (aculei pl.) A prickle, a small sharp point; specifically an ovipositor, especially when sting like as in Hymenoptera.

Agar: A gelatin-like material obtained from seaweed and used to prepare culture media on which microorganisms are grown and studied.

Aedeagus: In male insects, the penis or intromittent organ, situated below the scaphium and enclosed in a sheath.

Aerobic: A microorganism that lives or a process that occurs in the presence of molecular oxygen.

Aestivation: Dormancy in summer or during periods of continued high temperatures, or during a dry season.

Acrostichal setae: Very short hairs between the dorsocentral bristles.

Alare: Relating to the wings or having wings.

Alatae: Winged forms of aphids or aphids with wings.

Albedo: White or whiteness – reflective power, reflected light. The spongy white tissue on the inside of the rind of citrus fruit.

Alula: A lobe at the base of the wing especially in Diptera (flies).

Anal comb: The anal comb is a hard, comb-like appendage found on the last abdominal segment (Lepidoptera).

Anamorph: The imperfect or asexual stage of a fungus.

Anemotrophic/Anemotropism: Reaction to currents of air or wind movement.

Anepisternum: The upper division of the episternum when it is divided by a suture into two parts.

Antennae (pl. for Antenna): The paired segmented sensory organs, borne one on each side of the head may be referred to as horns or feelers.

Aperture: An opening, a hole, a gap.

Appressorium: The swollen tip of a hypha or germ tube that facilitates attachment and penetration of the host by a fungus or a parasitic plant.

Apodeme: Any one of the rigid processes forming the insect endoskeleton; a ridge or growth of the cuticle, serving for the attachment of muscles; an invagination of the body-wall; any lateral thoracic projection of an insect endoskeleton; also applied to any of the three types of in growths of the thorax.

Apterae (Aptera): Insects that have no wings.

Arista: A large bristle, usually located on the apical antennal segment (Diptera: flies).

Asci (pl. of Ascus): In ascomycetous fungi; a sac in which spores (usually eight) are produced and meiosis occurs.

Ascomata: Sexual fruiting body of an ascomycetous fungus that produces asci and ascospores; e.g. apothecium, ascostroma, cleistothecium, perithecium, pseudothecium.

Ascospores: Any of the spores in an ascus.

Aseptic: Free from disease producing microorganisms.

Asperites: Surface roughening or dot-like elevations.

Attractant: Something that attracts, such as a chemical pheromone that attracts insects.

Bacterium (pl. bacteria): A prokaryotic, microscopic, single-celled organism with a cell wall that increases by binary fission.

Binucleate: With two nuclei per cell.

Carina (pl. carinae): An elevated ridge or keel, not necessarily high or acute.

Chemotrophic: Reaction to chemical stimuli smell and taste; turn or bend under the influence of chemical substances.

Chorion: The outer shell or covering of the insect egg.

Chlorotic: Abnormal condition of plants in which the green parts lose their color or turn yellow as a result of chlorophyll production due to disease or lack of light.

Circulus: In coccids, called the ventralatia of MacGillivary; a glandular structure, the contents of which are discharged internally.

Cirrhus (pl. cirrhi): A curled, tendril-like mass of exuded fungal spores, held together by a slimy matrix

Cisanal setae: In coccids, the shorter and further two of the four setae (commonly known as hairs) near the caudal ring.

Clade: A monophyletic group.

Claw: A hollow sharp, multicellar organ, generally paired, at the end of the insect leg or one or more corneous sharp structures, which are the lobes of the maxillae.

Clavate: Club like, thickening gradually toward the tip.

Clypeus: That part of the head of the insect below the front, to which the labrum is attached anteriorly: in Diptera, often visible below the margin of the mouth in front, as a more or less visor-shaped piece.

Cocoon: A silken case inside which the pupa is formed.

Colonies (pl. for colony): A group of similar plants or animals living or growing together. For microorganisms, growth of a microorganism in mass, especially as a pure culture

Colonize: To infect and ramify through plant tissue with the growth of a pathogen

Colonization: Establishment and ramification of a pathogen within a host plant

Columella: A little rod, pillar or central axis.

Concave: Hollowed out; the interior of a sphere as opposed to the outer or convex surface.

Conidia: Asexual fungal spores.

Conidiogenous cells: Cells that produce and bear conidia

Convex: The outer curved surface of a segment of a sphere, opposed to concave.

Copious: Very plentiful, abundant.

Corolla: The second envelope of a flower usually composed of colored, leaf-like organs (petals), which may be united by their edges either in the basal part or throughout.

Costa: Any elevated ridge that is rounded at its crest. The thickened anterior margin of any wing, but usually the fore wings of an insect.

Coxae (pl. of Coxa): The basal segment of the leg of an insect, by means of which it is articulated to the body.

Crateriform: Like a shallow funnel or deep bowl; applied to depressions.

Crochets: The curved spines or hooks on the prolegs of caterpillars and on the cremaster of pupae.

Cross-fertilizing: To fertilize or be fertilized from another plant or animal by mutual exchanges of sperm between individuals.

Crown: The top of the head in Lepidoptera.

Cruciform: Cross-shaped.

Cubital cells: The wing area between the cubitus and anal vein; in the plural all the cells bounded anteriorly by the cubitus or its branches.

Cultivars: A variety of a plant species originating and continuing in cultivation and given a name in a modern language.

Cuticle: The noncellular outer layer of the body wall of an arthropod.

Cyme: A flat-topped or convex flower cluster with the central flowers blooming first.

Deflexed: Bent downward.

Defoliation: Loss of leaves from a plant, whether normal or premature

Denticle: A small tooth; in coccids a single tooth near the middle of the ventral aspect of the claw.

Deoxyribonucleic acid (DNA): the double-stranded, helical molecule that contains genetic code information; each repeating unit, or nucleotide, is composed of deoxyribose (a sugar), a phosphate group, and a purine (adenine or guanine) or a pyrimidine (thymine or cytosine) base.

Diapause: A period of arrested development and reduced metabolic rate, during which growth, differentiation, and metamorphosis cease, a period of dormancy not immediately referable to adverse environmental conditions.

Digitules: Appendages of the feet of Coccidae, which may be either broadly dilated or knobbed hairs; tenent hairs, empodial hairs.

Dicotyledonous (dicots): A flowering plant with two seed leaves characterized by embryos with two cotyledons, net veined leaves, flower parts in fours or fives.

Diffuse: Spread out or dispersed; not concentrated.

Dimorphic: Occurring in two distinct forms, differently colored in the two sexes.

Disinfest: To kill pathogens that have not yet initiated disease, or other contaminating microoganisms, that occur in or on inanimate objects such as soil or tools, or that occur on the surface of plant parts such as seed.

Dissected: Cut into parts; consisting of many lobes or segments, as some leaves.

Dissemination: To scatter seed, disseminate to scatter far and wide. For diseases, the spread of infectious material (inoculum) from diseased plants to healthy plants

Distal: Near or toward the free end of any appendage, that part of a segment farthest from the body.

Dorsal: Top or uppermost, pertaining to the upper side.

Ductus bursae: The duct in female Lepidoptera extending from the ostium to the bursa copulatrix.

Ecdyses: Molting; the process of shedding the exoskeleton.

Eclosion: Hatching from the egg.

Ectoparasite: A parasite that lives on the outside of its host.

Eggs: The oval or round body laid by a female, insect containing the germ of a new individual along with food for its development and having an enclosing shell or membrane.

ELISA (Enzyme-Linked ImmunoSorbent Assay): A serological test in which the sensitivity of the reaction is increased by attaching an enzyme that produces a colored product to one of the reactants

Endoparasite: Parasitic organism that lives and feeds from inside its host.

Entire: Whole not lacking any parts.

Epandrium: The ninth abdominal tergite of the male insect.

Epidemic: Prevalent and spreading rapidly among many individuals in a community at the same time.

Epidermis: The cellular layer of the skin; secreting the cuticula of insects

Elytra: The anterior leathery or chitinous wings of beetles.

Estivate: To pass the summer in a dormant state such as snails.

Eversible: Can be turned inside out

Exarate: Sculptured.

Exudate: Matter exuded – to ooze, discharge, to diffuse or seem to radiate.

Exuviae: The cast skin of an arthropod.

Eye Spots: Spots of color that look like eyes; usually on the wings of butterflies and moths.

Facultative: Capable of changing life-style.

Facultative diapause: May or may not need to diapause – not required for development.

Facultative parasite: A parasite, able to live as a saprotroph or a parasite.

Fecundity: The number of offspring per number of potential offspring (e.g. eggs).

Fastidious: Not easy to please, very critical or discriminating.

Femora (pl. for Femur): The third leg segment, located between the trochanter and the tibia.

Femur: Singular for femora; refer to femora.

Flagellum (pl: flagella): Hairlike, whiplike, or tinsel-like appendage of a motile cell, bacterium or zoospore that provides locomotion.

Flagellate: Having a flagellum or flagella.

Flagellomere: A segment of the antennal flaggellum. Male acuelate Hymenoptera have eleven flagellomeres, females have ten

Flange-like: Like a projecting rim or collar on a wheel, pipe, rail etc.

Flocculent: Containing downy or flaky masses.

Flush (citrus): New growth

Foraging: The act of searching for food and provisions.

Forewings: The anterior wings of an insect.

Frass: Plant fragments made by a wood-boring insect usually mixed with excrement; solid larval insect excrement.

Frons: The head sclerite bounded by the frontal (or frontogenal) and epistomal sutures and including the median ocellus.

Fungus: Any of the larger division of the thallophyte, including molds, mildews, mushrooms, rusts and smuts which are parasites on living organisms or feed on dead organic material, lacking chlorophyll, true roots, stems and leaves and reproduce by means of spores.

Furrow: A narrow groove made in another substance that resembles a deep narrow rut made by a wheel.

Gall: An abnormal swelling or localized outgrowth, often roughly spherical, produced by a plant as a result of attack by a fungus, bacterium, nematode, insect, or other organism

Gallery: Insect tunnel in bark and wood

Gaster: The most posterior body section of Hymenoptera, which consists of all abdominal segments except the first. The first abdominal segment is fused to the thorax and forms the propodeum.

Gena: The cheek; that part of the head on each side below the eyes, extending to the gular suture; in Odonata the area between the eyes and clypeus and mouth parts; in Diptera the space between the lower border of the eye and oval margin, merging into the face at the front and limited by the occipital margin behind.

Genua (pl. for genu): The knee.

Glabrous: Without hairs.

Globose: Spherical, perfectly round in all directions.

Graft: A shoot or bud of one plant or tree inserted or to be inserted into the stem or trunk of another where it continues to grow becoming a permanent part of the tree.

Gram-negative: Bacteria (cell wall) staining red or pink in the Gram staining procedure after treatment with Gram's stain

Gram-positive: Bacteria (cell wall) staining violet or purple in the Gram staining procedure after treatment with Gram's stain.

Gram stain: Procedure used for identification of bacteria in which crystal violet stain, Gram's iodine, ethyl alcohol and safranin stain are applied in succession to cells of the bacteria.

Gum: Gelatinous, sugary aggregate that is synthesized and exuded by plant tissues

Haustellum: The sucker; it is formed by the assemblage of inflexible setae, and inclosed in a rostellum or proboscis.

Haustorium (pl. haustoria): Specialized branch of a parasite formed inside host cells to absorb nutrients.

Head capsule: The welded together sclerites of the head which form a hard compact case for insects.

Helix: Spiral form.

Hemispherical: Shaped like the half of a globe or sphere.

Hermaphrodite: An individual in which the characters of both sexes are combined as in snails.

Honey dew: A sweetish excretion produced by certain insects notably aphids and coccids; also an exudates from the surface of some galls.

Humeral: Pertaining to the shoulder; located in the anterior basal portion of the wing.

Hyaline: Like glass, transparent colorless.

Hypandrium: The tenth sternite in mayflies, modified into a transverse plate; the ninth abdominal sternite of the male insect.

Hyphae (pl. of Hypha): Any of the thread-like parts making up the mycelium of a fungus.

Inclinate: Leaning or inclining.

Incubate: To brood; to cause to develop, as in an egg; cause to develop or take form. To keep (eggs, embryos, bacteria, etc.) in a favorable environment for hatching or developing.

Incubation period: The time between penetration of a host by a pathogen and the first appearance of disease symptoms.

Infection: Process in which an organism enters, invades, or penetrates and establishes a parasitic relationship with a host plant.

Infection peg: The specialized, narrow, hyphal strand on the underside of an appressorium that penetrates host cells

Inoculum: Pathogen or its parts, capable of causing infection when transferred to a favorable location.

Instar: The period or stage between molts in the larva, numbered to designate the various periods; e.g., the first instar is the stage between the egg and the first molt.

Integument: The outer covering or cuticle of the insect body.

Internodal area: The area that is that part of a plant stem between two successive nodes.

Interveinal: Between (leaf) veins

Isolate: (n.) A culture or subpopulation of a microorganism separated from its parent population and maintained in some sort of controlled circumstance;
(v.) To remove from soil or host material and grow in pure culture

Katepisternum: The lower part of the episternum.

Keel: An elevated ridge or carina.

Lanceolate: Lance or spear shaped, oblong tapering to the end.

Larvae (pl. for larva): An early, free living immature form of any animal that changes structurally when it becomes an adult usually by complex metamorphosis.

Latent period: The time between infection and the production of new inoculum; the time after a vector has acquired a pathogen and before it can be transmitted.

Lateral: Relating, pertaining, or attached to the side.

Lesions: Localized diseased area or wound.

Lobate: With lobes; divided by deep, undulating and successive incisions.

Lobe: Any prominent rounded process or excrescence on a margin or structure; specifically, the rounded, tooth like processes on the margin of the abdominal segments; in coccids, the semi-oval projection of the pygidial fringe.

Lumen: The enclosed spaces or cavity of any hollow or vesicular organ or structure.

Lunate: Crescent shaped.

Mandible: The first pair of jaws in insects, stout and tooth-like in chewing insects, needle or sword-shaped in piercing-mouthed sucking insects; the lateral upper jaws of biting insects; in muscoid larvae, the mouth hooks.

Medial: Referring to or at the middle.

Medium(pl. media): A mixture of organic and/or inorganic chemical compounds and water that provides the nutrients needed for the growth of a microorganism *in vitro*; for higher plants, a mixture of fertilizers and other components in which a plant is growing

Mesonotum: The primitively upper surface of the second or middle thoracic ring.

Microhabitat/microclimate: Weather conditions on a small scale, e.g. at the surface of the plant or within a crop

Micropyle: A very small opening in the our coat of an ovule, through which the pollen tube penetrates; The corresponding opening in the developed seed; one of the minute openings in the insect egg, through which spermatozoa enter in fertilization.

Microtrichia: Minute, hair-like structures found on the wings of certain insects; they resembles small covering hairs, but the absence of basal articulation distinguishes them; fixed hairs; aculei; minute non-movable hairs formed from cuticle; may produce an impression of cloudiness or color.

Mimicry: Strictly, the resemblance of one animal to another not closely related animal, living in the same locality; often loosely used to denote also resemblance to plants and inanimate objects; Bastesian mimicry is where one or two similar species is distasteful, the other is not distasteful (the mimic); Mullerian mimicry is where both species are distasteful.

Mollicute: One of a group of prokaryotic organisms bounded by flexuous membranes and lacking cell walls (phytoplasmas and spiroplasmas).

Molt: A process of shedding the exoskeleton, ecdysis.

Monocular: Having one eye or one eye piece for a microscope.

Mononucleate: Having one nucleus per cell.

Monogyne: A term used to describe a colony of ants with a single queen.

Nectary: A nectar-secreting gland in a flower.

Nematode: Nonsegmented roundworm (animal), parasitic on plants or animals, or free living in soil or water

Non-septate: Not having septa.

Notch: Ident, cut, or nick.

Notopleural: In Diptera, a depression, more or less triangular, situated immediately before the transverse suture and behind the humeri.

Nucleus: Dense aggregation of proteinaceous matter and nucleic acid in cells, surrounded by a membrane; contains chromosomes and controls heredity.

Nymphs: The immature stage (following hatching) of an insect that does not have a pupal stage; the immature stage of Acari (mite) that has eight legs.

Obligate parasite: (syn. biotroph) Organism that can grow only as a parasite in association with its host plant and cannot be grown in artificial culture media

Occiput: The dorsal posterior part of the head, between the occipital and post occipital sutures.

Ocellar: Referring to area around the ocelli such as ocellar bristles, ocellar triangle etc.

Ocellus: A simple eye of an insect or other arthropod.

Omnivore: An organism that eats both plants and animals.

Opaque: Without any surface luster; not transparent.

Orbital: Relating to an orbit; specifically the orbit of the eye.

Ostioles: In Heteroptera, one of the lateral metasternal external openings of the stink gland, placed near the coax in the adult; paired and dorsal on the abdomen of the nymph.

Ostium bursae: Ostium is the external genitalic opening of female Lepidoptera. Bursae the opening of the bursa copulatrix in Lepidoptera, equivalent to the vulva of female insects having the genital opening on the eighth segment.

Oviposit (oviposition): To deposit or lay eggs or ova. The act of depositing eggs.

Ovipositor: A specialized organ for depositing eggs in female insects.

Papilla: Nipple-like projection; used to describe the tip of some sporangia and the localized wall thickenings on the inner surface of plant cell walls at sites penetrated by fungi

Papillate: Having papilla

Paraphysis: An elongate sterile cell or hypha present in some fruiting bodies of fungi.

Parietalia: The dorsal sclerites of the head between the frontal and occipital regions.

Pecten: A comb.

Pedicel: Small slender stalk; stalk bearing an individual flower, inflorescence, or spore.

Penetration: Act or power of penetrating; the depth to which something penetrates.

Perineal pattern: Fingerprint-like pattern formed by cuticular striae surrounding the vulva and anus of the mature *Meloidogyne* female.

Periphery: The circumference of outer margin.

Periphysis: A paraphysis which grows into the neck of a perithecium

Perithecium: The globular or flask-shaped ascocarp of the Pyrenomycetes, having an opening or pore (ostiole)

Persistent transmission: (syn. circulative transmission)-A type of virus transmission in which the virus is acquired and transmitted by the vector after relatively long feeding times and remains transmissible for a prolonged period while in association with its vector

Petiolate: Stalked or placed on a stalk.

Phallus: The intromittent organ of the male, in which the male gonopore is situated; the penis.

Phasmids: Any of various sticklike or leaflike insects including walking sticks and leaf insects.

Pheromone: A substance given off by one individual that causes a specific reaction by other individuals of the same species, such as sex attractants, alarm substances etc.

Phialides: End cell of a conidiophore with one or more open ends through which a basipetal succession of conidia develops

Phialoconidia: Conidium on which phialides are borne.

Phloem: The vascular tissue in vascular plants, that conducts and distributes sugars and other dissolved foods from the places the food is produced to the places the food is needed or stored.

Phloem-limited: Limited to the plant phloem. Can only survive in the phloem.

Phytophagous: Plant eating

Piceous: a dark brown color.

Pinaculum: In caterpillars, an enlarged seta-bearing papilla forming a flat plate.

Pistil: The entire female section of the flower, including the eggs, ovary, style, and stigma.

Pleomorphic: Occurring in various distinct forms. In terms of cells, having variation in the size and shape of cells or their nuclei.

Polygene: A term used to describe an ant colony with two or more queens.

Polymerase chain reaction (PCR): A technique used to amplify the number of copies of a specific region of DNA in order to produce enough of the DNA for use in various applications uch as identification and cloning

Polyphagous (Polyphagy): Eating many kinds of food.

Pores: Large isolated punctures; a minute impression that perforates the surface; any small round opening on the surface.

Positive sense RNA: RNA that can serve directly as messenger RNA (see negative sense RNA).

Postalar: Behind the wings in position.

Postocellar: In Hymenoptera, the region on the dorsal aspect of the head bounded by the ocellar furrow, the vertical furrows and the caudal margin of the head.

Post ocular: Back of or behind the eyes.

Postpronotal (Postpronotum): The posterior region of the pronotum.

Postsutural: Behind the transverse suture.

Prepupae (pl. of prepupa): A quiescent instar between the end of the larval period and the pupal period proper; an active non feeding stage in the larva of Holometabola, a full fed larva.

Prescutum: The first of the four pieces composing the dorsal part, or tergum, of a thoracic segment of an insect. It is usually small and inconspicuous.

Presutural: Situated immediately in front of the transverse suture.

Prolegs: Any process or appendage that serves the purpose of a leg; specifically the fleshy unjointed abdominal legs of caterpillars and certain saw-fly larvae; abdominal feet, false legs; the anterior leg of the mature insect.

Prothoracic plate: Dorsal part of 1st thoracic segment of, for exemple, lepidopterous larva with cuticle thickened, often coloured, and shield-shaped

Protonymph: Second instar of a mite.

Pseudothecium (pl. pseudothecia): Perithecium-like fruiting body containing asci and ascospores dispersed rather than in an organized hymenium; an ascostroma with a single locule or cavity and containing bitunicate asci

Ptilinial suture: The crescentic groove cutting accress the froms above the antennal base in Diptera where the ptilinium has been withdrawn.

Ptilinum: An inflatable organ capable of being thrust out through the frontal suture just above the root of antennae, at emergence from the pupae.

Pterostigma: A thickened opaque spot along the costal margin of the wing, near the wing tip (also called the stigma).

Pubescent: Downy, covered with short fine closely set hairs.

Pupae (pl. of pupa): The stage between the larva and adult in insects with complete metamorphosis, a nonfeeding and usually inactive stage.

Puparium: The thickened, hardened barrel-like larval skin within which the pupae is formed.

Pycnidium: (pl. pycnidia) -Asexual, globose or flask-shaped fruiting body of certain imperfect fungi producing conidia

Pygidial: Pertaining to the pygidium (pygidium – the last dorsal segment of the abdomen).

Pyriform: Pear shaped.

Quadrate: Four-sided.

Rachis (floral): Thorn or point.

Radial: Arranged like rays starting from a common center; of or pertaining to the radius or radial wing vein.

Reclinate: Reflexed; directed backward; e. g. the bristles in Diptera.

Resistant (n. resistance) Possessing properties that prevent or impede disease development (see susceptible)

Ribonucleic acid (RNA): Several nucleic acids composed of repeating units of ribose (a sugar), a phosphate group, and a purine (adenine or guanine) or a pyrimidine (uracil or cytosine) base; transcribed from DNA and involved in translation to proteins.

Rostrum: Beak or snout such as weevils.

Rogue: To remove and destroy individual plants that are diseased, infested by insects, or otherwise undesirable

Rotund: Round or rounded out; plump or stout.

Rudimentary: Undeveloped.

Rugosties: Surface wrinkling

Sclerite: A hardened body wall plate bounded by sutures or membranous areas.

Sclerotized: Hardened.

Scrobes: Grooves formed for the reception or concealment of an appendage.

Scutellum: A sclerite of the thoracic notum the mesoscutellum appearing as a more or less triangular sclerite behind the pronotum especially in Hemiptera.

Scutum: The middle division of a thoracic notum, just anterior to the scutellum.

Sectors: Longitudinal views in Odonata, which strike the principal veins at an angle and usually reach the apex or hind margin; they are radial, subnodal, principal, nodal, median, short, and upper and lower of triangle.

Sedentary: Not active; settled or remaining in one place.

Semilunar spots: In the form of a half crescent.

Sensilla (pl. for sensillum): A simple sense organ, or one of the structural units of a compound sense organ.

Septa (pl. of septum): A part that separates two cavities or two masses of tissue.

Septate: Having or divided by a septum.

Serrate: Saw like; with notched edges like the teeth of a saw.

Sessile: Not supported on a stem or footstalk, Imobile.

Setae: A bristle; commonly known as hairs.

Setulose: Bearing truncated or blunt setae.

Sexual dimorphism: Sexes are different in form or color in the same species; may be seasonal or geographic; male and female look different by color form etc.

Shot-hole: A symptom in which small diseased fragments of leaves fall off and leave small holes in their place.

Sign: Indication of disease from direct observation of a pathogen or its parts (see symptom)

Sieve tube: A longitudinal tube in the phloem of flowering plants, consisting of a connected series of individual cells and serving to conduct organic food materials through the plant.

Siphunculi (pl. for siphunculus): The instrument of suction in sucking insects.

Sooty mold: A sooty coating on foliage and fruit formed by the dark hyphae of fungi that live in the honeydew secreted by insects such as aphids, mealybugs, scales, and whiteflies.

Spatulate: Rounded and broad at the top, attenuate at base. Shaped like a spoon, with a narrow end at the base.

Spermathecae: The sac or reservoir in the female that receives the sperm during coition.

Spiculose: Spiny

Spiracles: A breathing pore; in the plural the lateral openings on the segments of the insect body through which air enters the trachaea.

Spire: The upper part of a spiral shell of a gastropod.

Spores: The reproductive unit of fungi consisting of one or more cells; it is analogous to the seed in green plants.

Sporulate: To produce spores.

Stomata: Pores on the underside of plant leaves that can be opened or closed to control gas exchange and water loss. Openings in the epidermis (usually of the leaf) that allow gas exchange.

Striae (pl. for stria): In general, any fine longitudinal impressed line; in Coleoptera a longitudinal depressed line or furrow, frequently punctured, extending from the base to the apex of the elytra; In Lepidoptera, a fine transverse line.

Stridulatory apparatus: In general one part is like a file-like area and the opposing one a scraper or rasp; two ridged or roughed surface to be rubbed against each other.

Stridulatory movement: In insects rubbing the two roughed surfaces together to make creaking, grating or hissing sound or noise.

Stipitate (pl. for stipes): The foot-stalk of the maxilla, bearing the movable parts; modified into a piercing structure in some Diptera and into a lever for flexing the proboscis in others; either a pair of forceps in the male genitalia of aculeate Hymenoptera, the sagittae.

Stylet: A small style or stiff process; one of the trophy in Diptera and Hemiptera; a median dorsal element in the shaft of the ovipositor, formed of the united second vulvulae.

Subglobose: Not quite spherical.

Sublateral: In Diptera bristles situated in a line with the intraalars but in front of the suture.

Subulate appendage: Awl-shaped.

Suffusion: A clouding, spreading of one shade over another.

Supraanal plate/shield: Above the anus, a triangular sclerite covering the anal cavity above; present in many insects, sometimes in one sex only, often in both.

Surface sterilization: Placing seeds or plant material in a sterilizing material, often a sodium hypochlorite (bleach) solution, to remove contaminating microorganisms that may be on the surface.

Surstylus (pl. for surstyli): Paired appendages of the ninth abdominal tergite.

Susceptible: Prone to develop disease when infected by a pathogen (see resistance)

Sutures (pl. of suture): A seam or impressed line indicating the division of the distinct parts of a body wall; the line of juncture of the elytra in Coleoptera or the tegmina or hemelytra in other orders

Symptoms: The external and internal reactions or alterations of a plant as a result of disease.

Systemic: Of or affecting the entire organism or bodyily system; any of a group of pesticides that are absorbed into the tissues of plants, which in consequence become poisonous to insects etc. that feed on them.

Tactile: Of or pertaining to the sense of touch; used for touching.

Tarsus: The leg segment immediately beyond the tibia, consisting of one or more segments or subdivisions.

Teliomorph: The sexual or so-called perfert growth stage or phase in fungi.

Termitaria: A nest, natural or artificial or a colony of termites.

Tergites (pl. for tergite): A dorsal sclerite or part of a segment, especially when such part consists of a single sclerite.

Testa: The hard outer covering or integument of a seed.

Testaceous: Bearing a test or hard covering.

Thermoregulation: The regulation of body temperature.

Thorax: The body region behind the head, which bears the legs and wings.

Tibia: The fourth segment of the leg, between the femur and tarsus.

Tibiae: Plural of tibia.

Tibiotarsal: (Organ) in Collembola, sac-like swelling and an enlarged hair on the inner face of the hind tibiotarsus. (Segment) the fused tibia and tarsus in some insects.

Tomentum: A form of pubescence composed of matted, wooly hair.

Transient: Not permanent, temporary staying only for a short time.

Translucent: Shining through; transparent, letting light pass but diffusing it so that objects on the other side cannot be clearly distinguished.

Trilocular: Having three cells or cavities.

Trinucleate: Having three nuclei per cell.

Trochanter: The second segment of the leg, between the coax and femur.

Trophallaxis: The regurgitation of food by one animal for another. It is most highly developed in social insects such as the ants, in which individual colony members store food in their crops and regularly exchange it with other colony members and larvae to form a sort of "communal stomach" for the hive.

Truncate: Cut off square at the end.

Tubercle: A little solid pimple or small button, in Sphecoidea rounded lobes of the dorsal lateral margin of the pronotum; in caterpillars, body structures of the character, sometimes bear setae.

Tuberculate: Covered or furnished with tubercles; formed like a tubercle.

Tyloses: An overgrowth of the protoplast of a parenchyma cell into an adjacent xylem vessel or tracheid.

Umbilicus: A naval; or naval like impression.

Undulatory: Having a wave like form or motion, undulating.

Unicellular: Having or consisting of a single cell.

Unicolorous: Of one color throughout.

Urogomphi: Fixed or mobile processes found on the terminal segments of certain larvae; variously termined styli, cerci, pseudocerci, corniculi.

Variegation: Varied in color, of several colors in indefinite patterns.

Vector: An insect or animal that is able to transmit a pathogen.

Vein clearing: Disappearance of green color in or around leaf veins (a common symptom associated with virus infection).

Venom: Toxin secreted by animals; secreted by certain snakes and poisonous insects.

Vertex: The top of the head between the eye, front and occiput; in bees, that part of the head adjacent to and occupied by the ocelli.

Virus: A submicroscopic,intracellular, obligate parasite consisting of a core of infectious nucleic acid (either RNA or DNA) usually surrounded by a protein coat.

Vitta (pl. vittae): A broad longitudinal stripe.

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