University of Nebraska - Lincoln

DigitalCommons@University of Nebraska - Lincoln

Distance Master of Science in Entomology Projects

Entomology, Department of

2019

Integrated Pest Management Manual for Greenhouses

Evy Z. Santiago

Evy Z Santiago
Integrated Pest Management Manual for Greenhouses
Spring 2019

Introduction

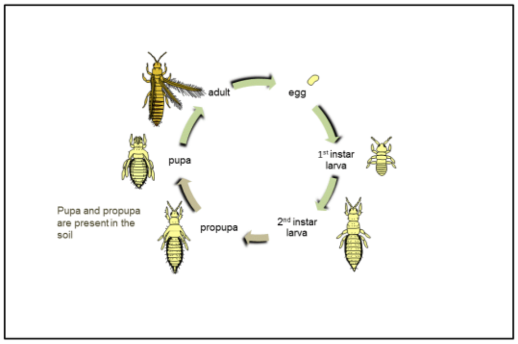
What is Integrated Pest Management?

The original definition of Integrated Pest Management (IPM) was introduced by Stern et al. in 1959 as Integrated Control. It was defined as applied pest control which combines and integrates biological and chemical control (Stern et al. 1959). Entomologists began exploring insect management alternatives to reduce reliance on insecticides in the 1950's after resistance and pest resurgence in agriculture was first noted. Today, the European Union follows a definition of IPM largely inspired from the definition given by the Food and Agriculture Organization of the United Nations (FAO) (Barzman et al. 2015). Integrated Pest Management means the careful consideration of all available pest control techniques and subsequent integration of appropriate measures that discourage the development of pest populations and keep pesticides and other interventions to levels that are economically justified and reduce or minimize risks to human health and the environment. IPM emphasizes the growth of a healthy crop with the least possible disruption to agro-ecosystems and encourages natural pest control mechanisms (FAO.org, 2019). The goal of IPM is a holistic and synergistic approach. It integrates preventive methods using different approaches. It focuses on biological, agronomic, mechanical, and physical principles, only resorting to selective pesticide usage when other tools are not successful (Barzman et al. 2015).

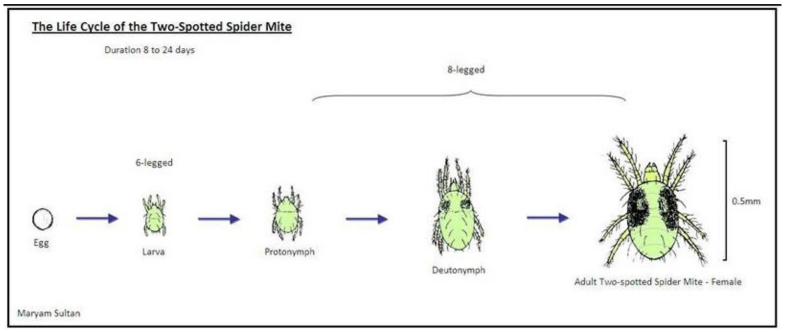
Greenhouses growing plants for research and development efforts have high plant densities with non-stop single crop production systems. This characteristic alone encourages the spread of pests. Furthermore, well fertilized and irrigated crops are often more sensitive to outbreaks of pests than outdoor crops (Gullino et al., 2002). While agronomic crops are the target of many pests, the main pests in a greenhouse environment are sucking pests such as whiteflies, aphids, thrips and mites. These are typically polyphagous insects, and are generally more problematic in greenhouses than in the field because of the ideal warm and moist protected environment, as well as the isolation from their natural enemies (Tian, 2000). These arthropods can develop and reproduce successfully throughout the year without hibernation under controlled conditions, producing more generations per year and causing more serious damage than under open field conditions (Tian, 2000). The greenhouse "acceptance threshold" is very low on agronomic crops with a R&D status. Nevertheless, integrated pest management within a controlled environment follows the same principles and management strategies to crops grown on fields. The following will be discussed: pest biology and identification, scouting and monitoring, cultural practices, biological control and chemical control.

Evy Z Santiago
Integrated Pest Management Manual for Greenhouses
Spring 2019

Common Greenhouse Pests Illustrations



Generalised thrips life cycle (©2019 Sonya Broughton)



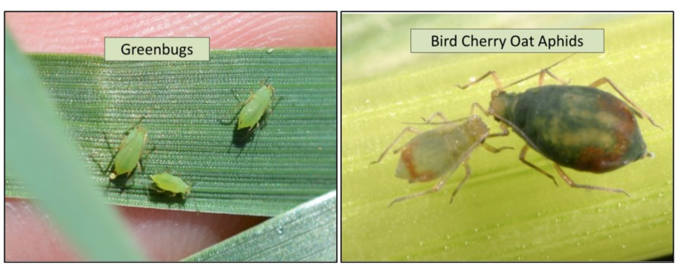
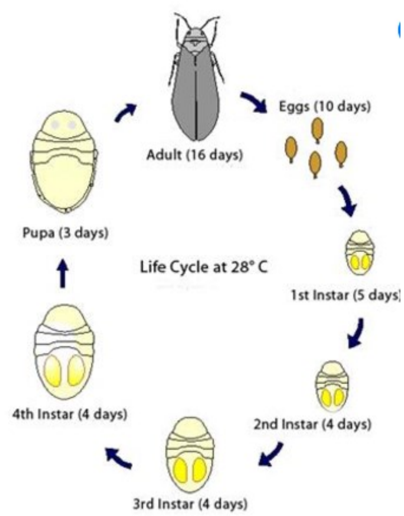


Photo credit: Dr. Jeff Whitworth and Dr. Holly Schwarting-Kansas State University



Common Greenhouse Pests and their Biology

Western flower thrips, *Frankliniella occidentalis* (Pergande)

Western flower thrips (WFT) are native to western North America. Their life cycle consists of an egg, 2 active feeding larval instars, 2 relatively quiescent pupal instars, and the adult. WFT are thigmotactic; adults and larvae aggregate in flowers or other concealed areas on plants, such as developing fruits, foliage, and floral buds (Reitz, 2009; Hansen et al. 2003).

Western flower thrips are an r-selected species (Pianka 1970; Reitz 2008). All studies of reproduction in western flower thrips have reported high fecundity for females. After an initial preoviposition period, a female can oviposit throughout her lifetime (Reitz 2008). Females deposit eggs under plant epidermis into leaves, petioles, flower bracts and petals, and developing fruit using a saw-like ovipositor (Reitz, 2009). With optimal temperatures and diets, females can produce up to 7 progenies per day and have average total lifetime fecundities exceeding 200 per female (Robb & Parrella 1991). In addition, females can live up to 45 days (Cloyd, 2010). This high level of fecundity leads to high intrinsic rates of population increase, so uncontrolled populations can multiply rapidly (Gaum et al. 1994; Gerin et al. 1994; Hulshof et al. 2003).

The breeding system of WFT is characterized by haplodiploidy (Ding et al. 2018; Reitz, 2009; Moritz, 1997). Arrhenotoky or fertilization of the eggs must be accomplished to produce a diploid female in contrast haploid males are produced from unfertilized eggs. (Moritz, 1997). This allows virgin females to survive and produce male offspring, which, in turn, build a bisexual cohort through oedipal mating (mating with their sons), (McCulloch et al. 2012). The life cycle of WFT takes two to three weeks to complete. To complete the life cycle between 7 to 13 days the temperature range must be between 26°C and 29°C (Cloyd, 2010). The first stadium is typically about half the length of the second (Gaum et al. 1994; Reitz 2008), after which feeding stops and pupation begins. The first pupal instar is termed the propupa, a non-feeding stage that is followed by the pupa, another non-feeding pupal stage. While WFT can remain on the host plant during pupation, they often drop to the soil to pupate. Emergence from the pupal stage takes 1 to 3 days (Broadbent et al. 2003; Buitenhuis & Shipp 2008). Under laboratory conditions, adult lifespan is relatively long compared with immature development time. For example, at 28°C, median egg to adult development time is 12 d, whereas median longevity for females is 26 d (Reitz 2008), with some females living up to 5 weeks (Trichilo & Leigh 1988; Hulshof & Vanninen 2002; Zhi et al. 2005; Reitz 2008).

Western flower thrips feed by piercing plant cells with their mouthparts and sucking out the contents (Hunter & Ullman 1989; Harrewijn et al. 1996). Adults and larvae feed in a similar manner, so both stages contribute to plant damage. Individuals tend to feed in localized areas, which results in silvered or necrotic patches on foliage, flowers and fruit. Feeding within developing buds leads to deformation of leaves or flowers (Childers, 1997). Western flower thrips females primarily feed on flower pollen, which may contain nutrients such as carbohydrates, proteins, sterols, and vitamins that enhance development rate and reproductive ability (Cloyd, 2010; Hulshof & Vanninen 2002; Hulshof et al. 2003; Zhi et al. 2005; Riley et al. 2007). Additionally, adults and larvae will prey on spider mite eggs (Trichilo & Leigh 1986).

Because of its polyphagous feeding and breeding behavior, WFT are exposed to a broad diversity of plant allelochemicals (Feyereisen 1999) hence must be able to metabolize a broad range of allelochemicals and respond to specific compounds by producing inducible enzymes (Li et al. 2007). Pesticide resistance studies have shown that WFT has various metabolic detoxification enzyme systems such as cytochrome P-450 monooxygenases, esterases, and glutathione S-transferases that could help it to overcome secondary plant defenses. It does seem that WFT has allelochemical-metabolizing genes that allow it cope with encountered allelochemicals (Li et al. 2007).

Two-spotted spider mite, *Tetranychus urticae* (Koch)

The two-spotted spider mite (TSSM) is a cosmopolitan and polyphagous mite affecting over 100 cultivated species such as cotton, corn, soybean, sunflowers, tomato, pepper, and numerous fruit and ornamental species (Bolland et al., 1998; Boom & Milstein, 2003).

Adults are small, ranging from 0.3 to 0.45 mm long, oval-shaped, and vary in color depending on the host plant from green- yellow to red-orange. Female TSSM are approximately 50% larger than males. They are broadly oval with 12 pairs of dorsal setae, whereas the male is slightly wedge-shaped with a narrow caudal end (Boudreaux and Dosse, 1963; Bolland et al., 1998). The two large dark spots on either side of the idiosoma, from which the species gets its common name can usually be seen through the body wall and they consist of food or bodily waste. Recently molted mites may not have these spots (Shih et al., 1976; English-Loeb, 1990).

The life cycle for TSSM consists of egg, larva, protonymph, deutonymph, and adult stages. The life cycle from egg to adult takes one to three weeks to complete, depending on ambient air temperature.

Adult females live about 30 days and can produce up to 200 eggs during a two-week period. The number and time to hatching vary depending on plant type and quality, based on nutritional status. Eggs are usually attached to fine silk webbing on the underside of leaves along the mid-veins and hatching takes about 3 days. After hatching, the first larval stage has three pairs of legs, while the protonymph, deutonymph and adult stages have four pairs. Spider mites are arrhenotokous, hence unmated females produce haploid eggs that develop into males while mated females produce diploid eggs that develop into females. When few males are in the population, females may mate with her sons. Regardless, inbreeding is minimal and spider mites have considerable genetic diversity which allows them to adapt to new conditions and environments (Helle and Sabelis, 1985; Sabelis, 1991; Krips et al., 1998; Tommaso et al., 2007). Females release sex pheromone at times causing some male mites to show guarding behavior during the female deutonymphs chrysalis stage. This behavior often end with one female mating with several males (Helle and Sabelis, 1985; Gotoh, 1997; Krips et al., 1998). The two-spotted spider mite can have 5 to 7 sequential generations per year, each generation developing within 15 to 28 days in favorable weather conditions (Helle & Sabelis, 1985).

In greenhouses, reproduction typically occurs year-round. Nevertheless, absence of host plants may provoke dormancy, causing mites to reside in cracks and crevices until new plant material is present. The length of diapause is determined by specific climatic conditions (Rotem and Agrawal, 2003; Kawakami et al., 2009). Females TSSM are consistently orange to orange-red during diapause (Shih et al., 1976; Veerman, 1985).

Two spotted spider mites prefer to feed on the undersides of leaves. They pierce individual plant cells, damaging the spongy mesophyll, palisade parenchyma, and chloroplasts. This decreases the chlorophyll content (55.26%) and even more dramatically the carotenoid content (79.3%) of the leaves (Hildebrand et al., 1986a). Therefore, the plant's ability to photosynthesize is affected and consequently, the plants are stunted. Damaged leaves appear bleached with yellow stains and stippled with small silvery-gray to yellowish speckles.

The two-spotted spider mite is equipped with glands located on the apex of their pedipalps that allow them to produce silk. This silk is used for web building, protection, dispersion and communication. When mite's population begins to increase on the undersides of leaves, the combined production of silk by the adults can serve as protection from some natural enemies and increase humidity levels preventing desiccation (Gerson, 1985). TSSM also use their silk for protection against pesticide residue, by spinning silk down avoiding contact with the insecticide (Margolies and Kennedy, 1988). The silks can also create a "roof" preventing pesticides to reach the leaf surface where mites are located. Silks can also help with dispersion especially when overcrowding occurs; at the apex of the plants mites can be picked up by the wind or animals for dispersal (Gerson, 1985, Clotuche et al., 2011).

Silk production can benefit some specialist predators such as phytoseiid mites (family Phytoseiidae) and ladybird beetles (van de Vrie et al., 1972) by signaling the presence of TSSM.

Greenbug aphid, *Schizaphis graminum* (Rondani)

The greenbug *Schizaphis graminum* is an important pest host of more than 70 grasses and cereals (Royer et al. 2015).

Greenbug adults and nymphs are oval-shaped with light-green bodies and a dark green dorsal line. Their antennae are dark and the legs are green with dark tarsi. The two cornicles located in the posterior end of the abdomen have dark tips. Alates have a brownish-yellow head and prothorax with black lobes on the thorax and a yellowish-green to dark-green abdomen and transparent wings that extend past the abdomen. Greenbugs develop through four nymphal stages, completed during a period of about one week under favorable conditions (Metcalf and Metcalf, 1993). The lower development threshold is approximately 3.5–6.0°C and upper development temperature is approximately 37°C (Kirkland et al. 1981, Walgenbach et al. 1988). Reproduction is mainly by apomictic parthenogenesis; all wingless greenbugs are female and give birth to live young. Alataes may be parthenogenetic females or male and female sexual morphs that can mate and produce eggs (Royer et al. 2015). Winged and wingless green-bugs measure 1.3–2.1 mm (Blackman and Eastop 2000). Per Washburn (1908) alatae sexual forms show dimorphism; males are slightly smaller than females.

Adults have a high reproductive capacity and population increases rapidly under favorable abiotic factors when there are no biotic factors present (Starks and Burton 1977).

Greenbug survival, growth, and reproduction are influenced significantly by temperature (Kirkland et al. 1981, Walgenbach et al. 1988). Jones et al. (2008) found that greenbugs supercool at approximately -26°C and winter survival would therefore not be possible in geographical areas with this temperature (Jones et al. 2008, Royer et al.2015).

Under optimal temperatures of 24–30°C, a newborn nymph will attain adulthood in 5 d, begin giving birth to live young in less than 0.5 d, and live for 25 d. One female can produce 60–80 offspring (Walgenbach et al. 1988). Under ideal conditions, numbers could double every 2 d; however, greenbug population abundance fluctuates because of mortality and emigration influenced by weather, natural enemies, and host plant conditions (Rogers et al. 1972, Wallin and Loonan 1977, Hamilton et al. 1982, Summy and Gilstrap 1983, Sumner et al. 1986, Kieckhefer and Gellner 1988, Royer et al. 2015). Greenbug, *Schizaphis graminum* feed by piercing and sucking on aboveground stems and leaves inhibiting plant growth or killing the plants which results in less yield and lower economic return (Stone et al. 2000, Burton et al. 1985, Royer et al. 2004, Royer et al. 2015). Greenbugs use needle-like, piercing-sucking mouthparts to feed on the phloem and inject toxic salivary enzymes that induce chlorosis around the feeding site and enhance food uptake (Pendleton et al. 2009, Royer et al. 2015). Further it reduces shoot and root biomass and tillering in wheat that persist throughout the entire growing season and may cause yield reductions (Burton 1986, Yoder et al. 2015). The injury caused by feeding during early stages appears as yellow or orange spots that have a dark necrotic lesion in the center in wheat and more red in sorghum (Yoder et al. 2015).

Bird cherry-oat aphid, *Rhopalosiphum padi* (Linnaeus)

The bird cherry-oat aphid is about 1.2-2.4 mm in length, soft bodied, and pear shaped. It is dark olive green with a characteristic reddish-brown patch on its back surrounding the base of the cornicles (Blackman & Eastop, 1984). The tips of legs, antennae, and cornicles are black and of average length. Adults may be winged or wingless. Winged forms are darker and develop under stressful conditions such as unfavorable weather, overcrowding, and reduction in food quality, allowing them to migrate over longer distances in search of more favorable host plants (Whitworth and Ahmad, 2008). Winged females appear in the third generation and then fly to the secondary hosts. These consist of a large number of species of Gramineae, Juncaceae and Cyperaceae (Rautapaa, 1970) on which parthenogenetic populations develop during the summer (Dixon & Glen, 1971).

The bird cherry-oat aphid complete life cycle is achieved on two different hosts. Sexual reproduction and cold-resistant egg production occurs in the fall on a primary host, from which females hatch early in spring producing two generations parthenogenetically (J.C. Simon et al. 1996).

The rapid development and asexual reproduction allows the bird cherry-oat aphid to reach tremendous population densities in a short time.

Bird cherry-oat aphid feed on phloem using needle-like piercing-sucking mouthparts and piercing the leaf, leaf sheath, or stem tissues near or just below the soil (Miller & Rasochova, 1997). It attacks all small grains including wheat, barley, oats, rye, and triticale. It may also be found on sorghum and corn. Heavy populations may cause a golden yellow streaking on the leaves. Occasionally heavy populations cause the flag to curl up in a tight corkscrew fashion that may trap the awns, resulting in a fish-hook appearance to the head. Additionally, heavy populations can excrete large amounts of honeydew promoting sooty mold and therefore reducing photosynthesis. More importantly, Quisenberry and Ni 2007 point out that cereal aphids, such as the bird cherry-oat aphid can decrease root proliferation affecting the size and yield in cereals in the absence of viruses, without any other visible symptom.

Whiteflies, *Bemisia tabaci* (Gennadius)

B. tabaci has a worldwide distribution, inhabiting every continent (Ghahari et al., 2013; CABI, 2017a). It is absent only in the most northern regions of Europe and Asia. This whitefly also is polyphagous, estimated to feed on over 900 host plants (McKenzie et al., 2014). It is becoming clear that *Bemisia tabaci* is a complex of morphologically indistinguishable species (Dinsdale et al., 2010; Tay et al., 2012).

Whiteflies are arrhenotokous; unfertilized eggs are haploid males and fertilized eggs are diploid females. The life cycle consists of the egg, four nymphal instars, and the adult stage (Walker et al., 2010). The egg stage has a pedicel that is inserted directly into epidermal tissue which absorbs water and possibly nutrients from the leaf (Walker et al., 2010). The eggs are arranged on the leaf surface in a partial egg circle (Naranjo et al., 2010).

Nymphs have an oval shape and are dorsoventrally flattened (Wakil et al. 2018). The first nymphal instar is highly mobile and his lateral margins have many setae. During this stage the nymph probes the leaf surface for a suitable location to insert its stylets into the phloem (Walker et al., 2010). Later instars will maintain this location and feed on the plant (Gill, 1990).

Adult *Bemisia tabaci* have wings that are held roof like over the abdomen and are covered in a white powder wax (Gill, 1990). Whiteflies show dimorphism; females are larger than the males and have a rounded abdomen. The male's abdomen is more pointed (Wakil et al. 2018).

Scouting and Monitoring Methods

A greenhouse crop scouting program is important to detect early infestations and in developing a successful IPM program. Scouting should be done once a week, and more often when an infestation is detected. Regular scouting and flagging every location scouted where insects have been found is also necessary to monitor the efficacy of control measures. Scouting can be done through visual inspection as well as sticky traps cards for catching flying insects. Sticky cards will help capture insect pests such as whiteflies, thrips and winged aphids.

Whiteflies, *Bemisia tabaci* (Gennadius)

It is suggested that a yellow sticky card (24 cm × 20 cm) could trap whitefly efficiently when placed vertically among plants and one card per 3–5 m, keeping the upper edge of the card at an equal level or slightly above the plants (Qiu and Ren 2006; Chen et al. 2012). Wang and Yang, 2017 placed five yellow sticky cards to monitor whiteflies in every 667 m^2 field using 5-points or "Z"-shaped methods, and replaced with new ones every 10 days. If the number of adult whitefly captured on the cards reached 0.25–0.5 adult/cm^2, biological control measures should be taken; if it reached 3–4 adult/cm^2, chemical control measures should be taken to suppress the outbreak of *B. tabaci* (Ren et al. 2014).

Western flower thrips, *Frankliniella occidentalis* (Pergande)

Western flower thrips (WFT) feed by piercing plant cells with their mouthparts and sucking out the contents (Hunter & Ullman 1989; Harrewijn et al. 1996). Adults and larvae feed in a similar manner, so both stages contribute to plant damage. Individuals tend to feed in localized areas, which results in silvered or necrotic patches on foliage, flowers and fruit. Feeding within developing buds leads to deformation of leaves or flowers (Childers 1997). Lightly blowing on blossoms and growing points aids in visual inspection as it causes thrips to become mobile, apparently because of the carbon dioxide contained in exhalation (Greer and Diver, 2000).

A hand lens is a useful tool to detect live thrips as well as signs of thrips activity such as black feces and silvery, flecked areas on leaves.

Monitoring of WFT is based on visual (colors, shape and size) or olfactory (host odor) signals which are primary cues used for host finding (Otieno et al. 2018). As a flower feeder, the canopy-dwelling WFT use color and scent cues for detection and orientation to their host plants (de Kogel and Koschier 2002). Abdullah et al. 2015, has shown that the use of blue sticky traps can significantly increase catches of *Frankliniella* spp. when enhanced with a stapled sachet impregnated with the kairomone (S)-(−)-verbenone. Other volatiles commonly used as attractants are currently commercially available. Furthermore, the use of blue (445nm) narrow-bandwidth LED lights has shown to work synergistically with blue sticky traps that have been enhanced with plant volatiles, increasing capture 2-fold (Otieno et al. 2018).

Aphids

Greenbug aphid, *Schizaphis graminum* (Rondani)

Bird cherry-oat aphid, *Rhopalosiphum padi* (Linnaeus)

Scouting and monitoring for aphids must follow a thorough visual inspection. Greenbugs and the bird cherry-oat aphid use needle-like, piercing-sucking mouthparts to extract plant sap from the phloem of their host. As they feed, they inject saliva into the plants to enhance food uptake. The greenbug feeding imparts visible injury to wheat and sorghum. Early damage in wheat appears as yellow or orange spots that have a dark necrotic lesion in the center. Sorghum reacts similarly, but the damaged spots first appear more reddish and then yellow. Feeding can result in death of sorghum leaves (Teetes and Johnson 1974) or death of seedling sorghum or wheat (Dahms and Wood 1957). The bird cherry-oat aphid (BCA), feeding damages cereals by depriving the plant of nutrients at their two-leaf stage, which causes 40–60% yield loss (Taheri et al. 2010). However, as mentioned previously BCA appears to cause no visible host plant damage because of feeding, although it may cause substantial yield loss (Leather et al. 1989, Riedell et al. 1999, Sahhed et al. 2007). Signs of an aphid infestation include honeydew or sooty mold on leaves, yellow spots on upper leaf surfaces, cast skins on leaves, curling of leaves, and distortion of new growth (Reddy, 2016). Knowing which species is infesting the crop is very important in successful detection and monitoring of aphid populations. (Reddy, 2016). Pay special attention to the undersides of leaves, stems, and new growth. Randomly selecting plants throughout the greenhouse, inspect undersides of leaves, buds, or tip growth and watch for honeydew and cast skins.

Yellow sticky cards placed horizontally at the top of the pot or container (if you are growing containerized plants) can be used for monitoring winged aphids. However, since winged aphids caught during the summer months may have blown in from the outdoors; sticky cards are not as reliable as visual inspections. Sticky cards are more useful in the winter months when aphids caught on the cards are not likely to have come in from the outside. It is better to rely primarily on visual inspections for aphid detection, and use sticky cards as a backup method.

Two-spotted spider mite, *Tetranychus urticae* (Koch)

A hand lens is a useful tool to see mites, especially by shaking plant foliage over a white piece of paper. However, heavy mite infestations are obvious to the naked eye on most crops, with leaf stippling and yellowing, obvious numbers of mites on the undersides of leaves, plus webbing.

According to Nihoul et al. 1991, the two-spotted spider mites prefer young leaves and their population in the upper part of the plant exhibits the potential of spreading. A newer way of scouting and monitoring for TSSM damage is using multispectral data. Hermann et al. 2011 was able to demonstrate damage separation on leaf tissue using the Green Normalized Difference Vegetation Index (GNDVI) and therefore able to monitor leaf damage with remote sensing tools.

On corn the first evidence of mite infestation can be noticed as yellow or white spotting on the surface of the leaf where the mites can be found feeding on the underside of the leaf. Use a hand lens to determine if there is stippling or webbing focusing on the underside of leaves or tap infested leaves over a white sheet of paper. Start scouting by examining the bottom leaves first and moving upwards, determining how far mites and symptoms have progressed up the plan. As infestations develop, leaf damage increases and photosynthesis decreases. Yield is affected when leaves at or above the ear level are infested. In the field spider mite control decisions are based on many factors including the mite species present, level of infestation, growth stage of the crop, cost of application and market price of the crop (Dhooria, 2016).

On soybean two-spotted spider mite feeding may cause leaf stippling, droppage, early senescence and pod shattering. A yield reduction of 40 to 60% has been reported during late vegetative and early reproductive stages (Dhooria, 2016; Cullen & Schramm, 2009).

On sorghum mite infested areas of leaves become a pale yellow initially and later the surface becomes a red-rust color. Dense webbings are found on undersurface of infested leaves. Mite populations generally increase after the emergence of sorghum grain heads. During early kernel development high mite populations can reduce fill grain. Insecticide applications should be considered when 30 % of leaves of most sorghum plants show mite damage symptoms (Dhooria, 2016).

Biological Control

Biological control is the periodic release of natural enemies such as parasitoids and predators in order to regulate or maintain insect or mite pest populations below damaging levels (Cloyd, 2012; Chambers et al. 1993; Gigon et al. 2016). There are various companies with commercially available biological control agents commonly used to manage insects pests in the greenhouse. Wollaeger et al. 2015, offers a free downloadable bulletin providing information on common greenhouse pests.

Aphids

Studies have shown that generalist predators present in high enough densities early in the growing season can suppress or contribute to the suppression of cereal aphid infestations (Chiverton, 1986; Edwards et al. 1979; Wratten and Powell, 1991; Brewer and Elliot, 2004).

Greenbugs are prey or hosts to numerous natural enemies (Brewer and Elliott 2004). Natural enemies include predators such as lady beetles (Coleoptera: Coccinellidae), lacewings (Neuroptera: Chrysopidae), damsel bugs (Hemiptera: Nabidae), spiders (Araneae), ground beetles (Coleoptera: Carabidae), syrphid flies (Diptera: Syrphidae) (Elliott et al. 2006; Royer et al. 2015), wasp parasitoids (Hymenoptera: Aphelinidae, Aphidiidae, Braconidae) (Walker et al. 1973, Archer et al. 1974, Gilstrap et al. 1984, Royer et al. 2015), and entomopathogenic fungi (Entomophthorales and Hypocreales) (Feng et al. 1990, Royer et al. 2015,).

Western Flower Thrips

Biological control of greenhouse thrips can be achieved through release of biocontrol agents such as predatory mites, lady beetles, and soil-dwelling mites.

An important biocontrol agent against WFT is *Orius insidiosus*, consuming more than 20 western flower thrips per day (Tommasini et al. 2004, Cloyd & Herrick, 2017)). Both nymphs and adults feed on a diversity of pests including: thrips, whiteflies, aphids, and spider mites in addition to plant sap and pollen in the absence of prey (Barber, 1936, Kiman et al. 1985, Cloyd & Herrick, 2017). Regulation of populations between WFT and the two-spotted spider mite, have been observed when both pest are present (Xu et al. 2006). *Orius* feeds on both larval and adult stages of western flower thrips (Kiman et al. 1985, Isenhour et al. 1985, van den Meiracker & Ramakers, 1991, Baez et al. 2004, Cloyd & Herrick, 2017).

Some important points about thrips biological control from Greer and Diver, 2000:

- A planned biocontrol release program that includes release before detection or as soon as thrips are found is necessary for the biocontrol to be effective.
- Adult female (*Neoseiulus*) predatory mites consume from 1 to 10 young thrips per day and have a 30-day lifespan. They can also survive on pollen and spider mites in the absence of thrips. A 1:2 ratio of predators to prey must be established. *Neoseiulus* attacks first instar only and does not move long distances from where it is first placed. They are most often applied in small piles at the base of plants, or in paper bags. Usually, a small hole is made in the bag, and mites move out of the bag slowly.
- Soil-dwelling *Hypoaspis,* a predaceous mites attack thrips found in the growing medium during their pre-pupal and pupal stages. *Hypoaspis* mites are usually applied only once per crop or season.
- *Thripobius semiluteus* is a parasitoid of greenhouse thrips nymphs.

Two-spotted spider mite

There are various commercially available predatory mites against the two-spotted spider mite. Those that shown most efficacy are in the family Phytoseiidae (Brust and Gotoh, 2017). There are many different species commercially available. The specialist phytoseiid mite *Phytoseiulus persimilis* is excellent during an outbreak as its population increases in response to surging spider mite populations (Nihoul, 1994; Drukker et al., 1997). However, it requires temperatures <80ºF and relative humidity between 60% to 80%. Other predatory mites include:

- *Mesoseiulus longipes,* which withstands warmer temperatures and a lower relative humidity than *P. persimilis.*
- *Neoseiulus californicus*, it can sustain longer without prey than *P. persimilis*
- *Neoseiulus fallacis*
- *Galendromus occidentalis*
- *Amblyseius andersoni*

The larvae of the predatory midge, *Feltiella acarisuga* also feeds on two-spotted spider mites.

Whiteflies

Encarsia formosa is the most successful natural enemy used for biocontrol of whiteflies in the world (Yang et al. 2014). The parasitic wasp kills whitefly nymphs in one of two ways: they either parasitize the nymph, or they kill the nymph right away and feed on its fluids. In addition to *E. formosa, Delphastus catalinae* has shown good efficacy under high density of whitefly nymphs (Zang & Liu, 2008). *D. catalinae* is a small ladybird beetle which feeds on all stages of whitefly, but prefers whitefly eggs and larvae. An adult beetle can consume up to 160 eggs or 12 whitefly larvae daily. The larva eats both whitefly larvae and about 1,000 whitefly eggs during its entire development. A single beetle can consume 10,000 whitefly eggs or 700 larvae during its lifetime (Biobest group, 2019). Other predators effective in whitefly biological control include the mites, *Amblydromalus limonicus* effective against whitefly eggs and larvae and thrips larvae, and *Amblyseius swirskii,* effective against whiteflies and western flower thrips.

Chemical Control

Chemical control applications must be initiated just after the accepted threshold has been reached. Decisions for chemical control are made based on: the efficacy on the pest, the mode of action, the method of application, potential phytotoxicity, environmental and personal safety, cost and restricted entry interval (REI).

Alternate chemical controls are the use of insecticidal soaps, botanical insecticides or natural pyrethrins and horticultural oils.

Insect growth regulators (IGRs) are another control option against pests. IGRs are placed into three general categories: juvenile hormone mimics or analogs; ecdysone antagonists; and chitin synthesis inhibitors. This complex mode of action prevents insects from rapidly developing resistance. IGRs generally do not have an effect on non-target species and most have minimal reentry restrictions (Reddy, 2016).

Microbial insecticides such as *Beauveria bassiana*, an entomopathogenic fungus used against aphids, mites, thrips, and whiteflies can also be an alternative to conventional chemical control. Strains of this entomopathogenic fungi provide good control of aphids. The conidia germinates on the surface of the insect and the hyphae penetrate the cuticle, the fungus consumes the internal contents of the host. *Beauveria bassiana* is available commercially for greenhouse ornamentals as Naturalis-O and for vegetables as BotaniGard.

Cultural Practices

The easiest and least expensive practice preventing pests from establishing in the greenhouse is sanitation. The greenhouse should be thoroughly cleaned and left empty for one week prior the beginning of the next crop. This enables removal of all pest stages, and starves any remaining adults. This strategy has worked for Western flower thrips. Shipp & Gillespie, 1993 found that western flower thrips would die at an air temperature of 104° F with a relative humidity of 10%. The goal of sanitation is to eliminate all possible sources of the pest. Per Hanan et al. 1978, Berlinger et al. 1999 and Weintraub et al. 2017, sanitation practices and guidelines include:

- Double access doors (interlocking) to limit the entrance of arthropods (and improve thermal insulation).
- Footbaths filled with disinfectants at points of access to the greenhouse, to reduce pathogen movement.
- Hand and implement washing stations supplied with soap and water for sanitizing before movement from one area to another. Reduce staff movement in and through areas that are known to have insect pest populations, visit these areas last and do not re- enter an uninfested area.
- Removal of weeds within and immediately surrounding the greenhouse that can be a source of both arthropod pests and pathogens.
- Removal and disposal of infested and infected plant material that may harbor pests and pathogens.

Another practice to prevent insect pests from entering the greenhouse is by placing screens on greenhouse openings including side and ridge vents. The usage of screens with high ultraviolet-absorbing properties has been demonstrated to be an effective barrier against the entrance of insects such as aphids, leafhoppers, thrips and whiteflies in the greenhouse (Antignus 2000, Weintraub et al. 2008, Kigathi and Poehling 2012, Legarrea et al. 2014).

Water and fertility management play important roles as cultural control. Excessive nitrogen fertilization will increase the number of phytophagous insects such as whitefly and aphids as well as exacerbate the honeydew production (Bi et al. 2001, 2005). Dry and hot weather favors mite reproduction and survival especially if accompanied by drought stress in the crop. Similar observations have been noted on *B. tabaci* populations on water-stressed cotton as compared to well-watered plants (Flint et al. 1996, Wang & Yang, 2017). Irrigations should be timed properly to avoid moisture stress.

Literature Cited

Antignus, Y. 2000. Manipulation of wavelength-dependent behaviour of insects: an IPM tool to impede insects and restrict epidemics of insect borne viruses. Virus Res. 71: 213–220.

Baez, I., Reitz, S.R. and Funderburk, J.E. 2004.Predation of *Orius insidiosus* (Heteroptera: Anthocoridae) on Species and Life Stages of *Frankliniella* Flower Thrips (Thysanoptera: Thripidae) in Pepper Flowers. Environmental Entomology, 33, 662-670.

Barber, G.W. 1936.*Orius insidiosus* (Say), An Important Natural Enemy of the Corn Earworm. United States Department of Agriculture Technical Bulletin, Washington DC, 504 p

Barzman M. et al. (2015) Research and Development Priorities in the Face of Climate Change and Rapidly Evolving Pests. In: Lichtfouse E. (eds) Sustainable Agriculture Reviews. Sustainable Agriculture Reviews, vol 17. Schweiz: Springer. pp.1-27

Bi J.L., Ballmer G.R., Hendrix D.L., Henneberry T.J., Toscano N.C. 2001. Effect of cotton nitrogen fertilization on *Bemisia argentifolii* populations and honeydew production. Entomol Exp Appl 99:25–36

Bi J.L., Lin D.M., Li K.S., Toscano N.C. 2005. Impact of cotton planting date and nitrogen fertilization on *Bemisia argentifolii* populations. Insect Sci 12:31–36

Blackman R. L. Eastop V. F. 2000. Aphids on the world's crops: An identification and information guide. John Wiley & Sons, West Sussex, England.

Bolland, H.R.; Gutierrez, J.; Flechtmann, C.H.W. 1998: *World catalogue of the spider mite family (Acari: Tetranychidae)*. Leiden; Boston; Köln: Brill

Boom, M.E.V.D. and Milstein, D. (2003): Cyclometalated phosphine-based pincer complexes: mechanistic insight in catalysis, coordination, and bond activation. Chem. Rev., 103, 1759–1792

Boudreaux, H.B., Dosse, G., 1963. Concerning the names of some common spider mites. Advances in Acarology 1, 350-364

Brewer, M. J. and N.C. Elliott. 2004. Biological Control of Cereal Aphids in North America and Mediating Effects of Host Plant and Habitat Manipulations
Annual Review of Entomology 2004 49:1, 219-242

Broadbent, A. B., Rhainds, M., Shipp , L., Murphy , G., and Wainman , L. 2003. Pupation behaviour of western flower thrips (Thysanoptera: Thripidae) on potted chrysanthemum. Canadian Entomol. 135: 741 -744.

Brust, G. E. and T. Gotoh 2017. Mites: Biology, Ecology, and Management. In: Sustainable Management of Arthropod Pests of Tomato (Wakil, W., Brust, G. E., and Perring, T. M. eds.). Elsevier Inc. (Amsterdam), pp. 111-130.

Buitenhuis, R., and Shipp, J. L. 2008. Influence of plant species and plant growth stage on Frankliniella occidentalis pupation behavior in greenhouse ornamentals. J. Appl. Entomol. 132: 86-88

Burton R.L. 1986. Effect of greenbug (Homoptera: Aphididae) damage on root and shoot biomass of wheat seedlings. *J. Econ. Entomol.* 79: 633–36

Burton R. L. Morrison R. D. Starks K. J. Simmon D. D. 1985. Seasonal damage by greenbugs (Homoptera: Aphididae) to a resistant and a susceptible variety of wheat. J. Econ. Entomol. 78: 395–401.

Childers, C. C. 1997. Feeding and oviposition injuries to plants, pp. 505-537, In T. Lewis [ed.], Thrips as Crop Pests. CAB International, New York.

Chiverton PA. 1986. Predator density manipulation and its effects on populations of *Rhopalosiphum padi* (Hom.: Aphididae) in spring barley. Ann. Appl. Biol. 109: 49– 60

Chow, A., Chau, A. and Heinz, K.M. 2008.Compatibility of *Orius insidiosus* (Hemiptera: Anthocoridae) with *Amblyseius (Iphiseius) degenerans* (Acari: Phytoseiidae) for Control of *Frankliniella occidentalis* (Thysanoptera: Thripidae) on Greenhouse Rose. Biological Control, 44, 259-270.

Clotuche, G., Mailleux, A., Fernandez, A.A., Deneubourg, J., Detrain, C., Hance, T., 2011. The formation of collective silk balls in the spider mite *Tetranychus urticae* Koch. PLoS One 6 (4), e18854

Cloyd, R.A. 2010. Western Flower Thrips Management on Greenhouse Grown Crops. K State University Fact sheet MF-2922. 8 pp.
https://www.bookstore.ksre.ksu.edu/pubs/mf2922.pdf

Cloyd, R.A. 2012. Chapter 13: Insect and Mite Management in Greenhouses. In: Nelson, P.V., Ed., Greenhouse Operation and Management, 7th Edition, Pearson Prentice Hall, Upper Saddle River, NJ, 391-414

Cloyd, R. and Herrick, N. 2017. Ecology and Role of the Rove Beetle, *Dalotia coriaria*, and Insidious Flower Bug, *Orius insidiosus*, in Greenhouse Biological Control Programs. *Advances in Entomology*, 5, 115-126.

Dhooria, M.S.(2016)Mite Pests of Field Crops. In: Fundamentals of Applied Acarology. Singapore: Springer pp. 275-305

Ding, T., Chi, H., Gökçe, A., Gao, Y., & Zhang, B. (2018). Demographic analysis of arrhenotokous parthenogenesis and bisexual reproduction of Frankliniella occidentalis (Pergande) (Thysanoptera: Thripidae). Scientific reports, 8(1), 3346.

Edwards CA, Sunderland KD, George KS. 1979. Studies on polyphagous predators of cereal aphids. J. Appl. Ecol. 16: 811– 23

English-Loeb, G. M. 1990. Plant drought stress and outbreaks of spider mites: a field test. Ecology, 71:1401-1411.

Feyereisen, R. 1999. Insect P450 enzymes. Annu. Rev.Entomol. 44: 507-533

Fransen, J.J. and Tolsma, J. (1992) Releases of the Minute Pirate Bug, *Orius insidiosus* (Say) (Hemiptera: Anthocoridae), Against Western Flower Thrips, *Frankliniella occidentalis* (Pergande) on Chrysanthemum. Mededelingen-Faculteit Landbouwkundige en Toegepaste Biologische Wetenschappen, Universiteit Gent, 57, 479-484.

Flint HM, Naranjo SE, Leggett JE, Henneberry TJ (1996) Cotton water stress, arthropod dynamics, and management of *Bemisia tabaci* (Homoptera: Aleyrodidae). J. Econ. Entomol. 89:1288–1300

Food and Agriculture Organization of the United Nations. 2019. AGP-Integrated Pest Management
http://www.fao.org/agriculture/crops/thematic-sitemap/theme/pests/ipm/en/

Funderburk, J., Stavisky, J. and Olson, S. 2000.Predation of *Frankliniella occidentalis* (Thysanoptera: Thripidae) in Field Peppers by *Orius insidiosus* (Hemiptera: Anthocoridae). Environmental Entomology, 29: 376-382.

Gaum, W. G., J. H. Giliomee, and K. L. Pringle. 1994. Life history and life tables of western flower thrips, *Frankliniella occidentalis* (Thysanoptera, Thripidae), on English cucumbers. Bull. Entomol. Res. 84: 219–224.

Gerin, C., Hance, T.H., and G. Van Impe. 1994. Demographical parameters of *Frankliniella occidentali*s (Pergande) (Thysanoptera, Thripidae). J. Appl. Entomol. 118: 370–377.

Gerin, C., Hance, T.H., and G. Van Impe. 1999. Impact of flowers on the demography of western flower thrips *Frankliniella occidentalis* (Thysanoptera: Thripidae). J. Appl. Entomol. 123: 569–574

Gerson, U., 1985. Webbing. In: Helle, W., Sabelis, M.W. (Eds.), Spider Mites: Their Biology, Natural Enemies and Control. Elsevier, Amsterdam, The Netherlands, pp. 223-231

Gigon, V., Camps, C. and Le Corff, J. 2016. Biological Control of *Tetranychus urticae* by *Phytoseiulus macropilis* and *Macrolophus pygmaeus* in Tomato Greenhouses. Experimental and Applied Acarology, 68, 55-70.

Gilrein, Dan. 1999. Control thrips from the start. Greenhouse Management and Production. January. p. 72–74

Gotoh, T. 1997. Annual life cycles of populations of the two-spotted spider mite, *Tetranychus urticae* Koch (Acari: Tetranychidae) in four Japanese pear orchards. Applied Entomology and Zoology 32, 207-216.

Greer, L., and S. Diver. 1999. Integrated Pest Management for Greenhouse Crops: Pest Management Systems Guide. Appropriate Technology Transfer for Rural Areas.
http://attra.ncat.org/attra-pub/download.php?id=48

Gullino, M.L., Albajes, R., van Lenteren, J.C., 2002. Setting the stage: characteristics of protected cultivation and tools for sustainable crop protection. In: Albajes, R., Gullino, M.L., van Lenteren, J.C., Elad, Y. (Eds), Integrated Pest and Disease Management in Greenhouse Crops. Kluwer Academic Publishers, New York, Boston, Dordrecht, London, Moscow, pp.1-15

Hansen, E. A., J. E. Funderburk, S. R. Reitz, S. Ramachandran, J. E. Eger, and H. McAuslane. 2003. Within-plant distribution of *Frankliniella* species (Thysanoptera: Thripidae) and *Orius insidiosus* (Heteroptera: Anthocoridae) in field pepper. Environ. Entomol. 32: 1035–1044.

Hamilton G. C. Kirkland R. L. Peries I. D. R. 1982. Population ecology of Schizaphis graminum (Rondani) (Homoptera: Aphididae) on grain sorghum in central Missouri. Environ. Entomol. 11: 618–628.

Harrewijn, P., Tjallingii, W. F., and Mollema, C., 1996. Electrical recording of plant penetration by western flower thrips. Entomol. Exp. Appl. 79: 345-353.

Helle, W., Sabelis, M.W. (Eds.) 1985. Spider Mites: Their Biology, Natural Enemies and Control. Elsevier, Amsterdam, The Netherlands, p.458

Hildebrand, D.F., Rodriguez, J.G., Brown, G.C., Luu, K.T., and Volden, C.S. 1986a. Peroxidative responses of leaves in two soybean genotypes injured by twospotted spider mites (Acari:Tetranychidae) Journal of Economic Entomology 79: 1459-1465.

Horowitz, A. R. and I. Ishaaya 2004. Biorational Insecticides – Mechanisms, Selectivity and Importance in Pest Management Programs. In: Insect Pest Management- Field and Protected Crops (A. R. Horowitz and I. Ishaaya, eds.), pp.1-28. Springer, Berlin, Heidelberg, New York

Hulshof, J., and L. Vanninen. 2002. Western flower thrips feeding on pollen, and its implications for control, pp. 173-179 In R. Marullo and L. A. Mound[eds.], Thrips and Tospoviruses: Proc.7th Intl. Symp.on Thysanoptera, July 2-7, Reggio Calabria, Italy. Australian National Insect Collection, Canberra

Hulshof, J., Ketoja, E. and L. Vanninen. 2003. Life history characteristics of *Frankliniella occidentalis* on cucumber leaves with and without supplemental food. Entomologia Experimentalis et Applicata 108: 19–32.

Hunter, W. B., AND Ullman, D. E. 1989. Analysis of mouthpart movements during feeding of *Frankliniella occidentalis* (Pergande) and *Frankliniella schultzei* Trybom (Thysanoptera: Thripidae). Int. J.Insect Morphol. Embryol. 18: 161-172.

Isenhour, D.J. and Yeargan, K.V. 1981. Predation by *Orius insidiosus* on the Soybean Thrips, *Sericothrips variabilis*: Effect of Prey Stage and Density. Environmental Entomology, 10, 496-500.

Jones D. B. Giles K. L. Elliott N. C. 2008. Supercooling points of Lysiphlebus testaceipes and its host Schizaphis graminum. Environ. Entomol. 37: 1063–1068.

Kawakami, Y., Numata, H., Ito, K., Gotoh, S.G. 2009. Dominant and recessive inheritance patterns of diapause in the two-spotted spider mite *Tetranychus urticae*. Journal of Heredity 101, 20-25

Kieckhefer R. W. Gellner J. L. 1988. Influence of plant growth stage on cereal aphid reproduction. Crop Sci. 28: 688–690.

Kigathi, R., and H.-M. Poehling. 2012. UV-absorbing films and nets affect the dispersal of western flower thrips, *Frankliniella occidentalis* (Thysanoptera: Thripidae). J. Appl. Entomol. 136: 761–771.

Kiman, Z.B. and Yeargan, K.V. 1985.Development and Reproduction of the Predator *Orius insidiosus* (Hemiptera: Anthocoridae) Reared on Diets of Selected Plant Material and Arthropod Prey. Annals of the Entomological Society of America, 78, 464-467.

Kirkland R. L. Peries K. D. Hamilton G. C.. 1981. Differentiation and developmental rate of nymphal instars of green bug reared on sorghum. J. Kans. Entomol. Soc. 54: 743–747

Krips, O. E., Witul, A., Willems, P. E. L. and Dicke, M. (1998) Intrinsic rate of population increase of the spider mite *Tetranychus urticae* on the ornamental crop gerbera: intraspecific variation in host plant and herbivore. Entomol. Exp. Appl. 89: 159–168

Legarrea, S., E. Velazquez, P. Aguado, A. Fereres, I. Morales, D. Rodriguez, P. Del Estal, and E. Vinuela. 2014. Effects of a photoselective greenhouse cover on the performance and host finding ability of *Aphidius ervi* in a lettuce crop. BioControl. 59: 265–278.

Li, X., Schuler, M. A., and Berenbaum, M. R. 2007.Molecular mechanisms of metabolic resistance to synthetic and natural xenobiotics. Annu. Rev. Entomol. 52: 231-253

Margolies, D.C., Kennedy, G.G. 1988. Fenvalerate-induced aerial dispersal by the twospotted spider mite. Entomologia Experimentalis et Applicata 46, 233-240

McCulloch, J. B., & Owen, J. P. (2012). Arrhenotoky and oedipal mating in the northern fowl mite (Ornithonyssus sylviarum) (Acari: Gamasida: Macronyssidae). *Parasites & vectors*, *5*, 281.

Moritz, G. (1997) Structure, growth and development. In Thrips As Crop Pests (Lewis, T., ed.), p. 49. CAB International, New York

Parella, Michael P. 1995. Thrips management guide — Part 1: Prevention and control. GrowerTalks. April. p. 30, 32, 34, 36, 38.

Pendleton B. P. Copland A. L. P. Michels G. J.Jr. 2009. Effect of biotype and temperature on fitness of greenbug (Hemiptera: Aphididae) on sorghum. J. Econ. Entomol. 102: 1624–1627.

Pendleton B. B., Palousek Copeland A. L., Michels G. J., 2009. Effect of biotype and temperature on fitness of greenbug (Hemiptera: Aphididae) on sorghum. Journal of Economic Entomology, 102: 1624-1627.

Pinka, E. R. 1970. On r and K selection. American Nat.104: 592-597.

Qiu B.L., Ren S.X. 2006.Using yellow sticky traps to inspect population dynamics of *Bemisia tabaci* and its parasitoids. Chin Bull Entomol 43(1):53–56. in Chinese, English abstract

Reddy, P.P. 2016.Thrips and their Management. In: Sustainable Crop Protection under Protected Cultivation, Singapore: Springer pp 213-220

Reitz, S. R. 2008. Comparative bionomics of *Frankliniella occidentalis* and *Frankliniella tritici.* Fla Entomol. 91: 474-476

Reitz, S. R. 2009. Biology and ecology of the western flower thrips (Thysanoptera: Thripidae): the making of a pest. Fla. Entomol. 92: 7–13.

Ren SX, Liu TX, Du YZ, Peng ZQ, Qiu BL, Ge F (2014) Systematic investigation and monitoring of whitefly pests in a vegetable ecosystem. Chin J Appl Entomol 51(3):859–862 (in Chinese, English abstract)

Riley, D. G., Chiturri, A., and Sparks JR, A. N. 2007.Does natural deposition of pine pollen affect the ovipositional behavior of *Frankliniella occidentalis* and *Frankliniella fusca*? Entomol. Exp. Appl. 124: 133-141

Robb, K. L. & M. P. Parrella. 1988. Chemical and non-chemical control of western flower thrips, pp. 94-103. In A. D. Ali, J. Hall & M. P. Parrella [eds.J, Proc. 4th Con. on Insect and Disease Management on Ornamentals, Kansas City, MO

Rogers C. E. Eikenbary R. D. Starks K. G.. 1972. A review of greenbug outbreaks and climatological deviations in Oklahoma. Environ. Entomol. 1: 664–668.

Rotem, K A and Agrawal A. A. (2003) Density dependent population growth of the two-spotted spider mite, *Tetranychus urticae*, on the host plant *Leonurus cardiaca*. Oikos 103: 559-565

Royer, T. A. Giles K. L. Kindler S. D. Elliott N. C. 2004. Cereal aphid expert system and Glance ‘n Go sampling for greenbugs: Questions and answers. CR-7191. Oklahoma Cooperative Extension Service, Stillwater, OK.

Royer, T.A., Pendleton, B.B., Elliott, N.C., Giles, K.L. 2015. Greenbug (Hemiptera: Aphididae) Biology, Ecology, and Management in Wheat and Sorghum, Journal of Integrated Pest Management, 6:1

Sabelis M W (1991) Life-history evolution of spider mites. In: Schuster R and Murphy P W (eds): The Acari.
Reproduction, Development and Life-History Strategies. Chapman & Hall, London, pp. 23–49.

Shih, C. T., Poe, S. L. & Cromroy, H. L. 1976: Biology, life table, and intrinsic rate of increase of Tetranychus urticae. Ann. Entomol. Soc. Am., 69: 362—364.

Shipp, J.L. and T.J. Gillespie. 1993. Influence of temperature and water vapor pressure deficit on survival of *Frankliniella occidentalis* (Thysanoptera: Thripidae). Environmental Entomology. August. p. 726-732

Starks K. J. Burton R. L. 1977. Preventing greenbug outbreaks. Report no. 309, USDASci. Educ. Admin. Leaflet. Washington, DC.

Stern V, Smith R, van den Bosch R, Hagen K. 1959. The integration of chemical and biological control of the spotted alfalfa aphid: The integrated control concept. Hilgardia 29(2):81-101

Stone BS, Shufran RA, Wilde GE (2000) Life history of multiple clones of insecticide resistant and susceptible greenbug *Schizaphis graminum* (Homoptera: Aphididae). J Econ Entomol 93:971–974

Summy K. R. Gilstrap F. E. 1983. Facultative production of alates by greenbug and corn leaf aphid, and implications in aphid population dynamics. J. Kans. Entomol. Soc. 56: 434–440.

Sumner L. C. Johnson J. C. McNew R. W. Eikenbary R. D. Dorschner K. W. Ryan J. D. 1986. Reproduction of *Schizaphis graminum* (Homoptera: Aphididae) on resistant and susceptible wheat genotypes during simulated drought stress induced with polyethylene glycol. Environ. Entomol. 15: 756–762.

Tian, Y.Q., 2000. Biological Control of Vegetable Pests. Jindun Press, Beijing

Tommasini, M.G., van Lenteren, J.C. and Burgio, G. 2004. Biological Traits and Predation Capacity of Four *Orius* Species on Two Prey Species. Bulletin of Insectology, 57, 79-93.

Tommaso A, Sílvia A and Josep J (2007) Estimating the intrinsic rate of increase of *Tetranychus urticae*: which is the minimum number of immature individuals to consider? Exp. Appl. Acarol. 41: 55-59

Trichilo, P. J., and Leigh, T. F. 1986. Predation on spider mite eggs by the western flower thrips, *Frankliniella occidentalis* (Thysanoptera: Thripidae), an opportunist in a cotton agroecosystem. Environ.Entomol. 15: 821-825

Trichilo, P. J., and Leigh, T. F. 1988. Influence of re-source quality on the reproductive fitness of flowerthrips (Thysanoptera: Thripidae). Ann. Entomol.Soc. America 81: 64-70

van den Meiracker, R.A.F. and Ramakers, P.M.J. (1991) Biological Control of the Western Flower Thrips *Frankliniella occidentalis* in Sweet Pepper with the Anthocorid Predator *Orius insidiosus*. Mededelingen-Faculteit Landbouwkundige en Toegepaste Biologische Wetenschappen, Universiteit Gent, 56, 241-249.

van Lenteren, J.C. (2012) The State of Commercial Augmentative Biological Control: Plenty of Natural Enemies, but a Frustrating Lack of Uptake. BioControl, 57, 1-20.

Veerman A. 1985. Diapause. In Spider mites: their biology, natural enemies and control, Vol. 1A, W.Helle and M.W.Sabelis (eds), Elsevier, Amsterdam. pp. 279–316.

Vrie, M. van de, McMurtry, J. A. & Huffaker, C. B. 1972. Ecology of tetranychid mites and their natural enemies: a review. III. Biology, ecology, and pest status, and host-plant relations of tetranychids. Hilgardia, 41: 343-432

Walgenbach D. D. Elliott N. C. Kieckhefer R. W. 1988. Constant and fluctuating temperature effects on developmental rates and life table statistics of the greenbug (Homoptera: Aphididae). J. Econ. Entomol. 81: 501–507.

Wallin J. R. Loonan D. V. 1977. Climatic factors affecting the development of high populations of greenbug in Texas and Oklahoma. Plant Dis. Rep. 61: 4–6.

Washburn F. L. 1908. Sexual forms of Toxoptera graminum Rond. Can. Entomol. 40: 53–54.

Weintraub, P., Recht, E., Mondaca, L., Harari, A.R., Diaz, B.M., and J. Bennison. 2017. Arthropod Pest Management in Organic Vegetable Greenhouses. Journal of Integrated Pest Management, 8(1): 29; 1–14

Weintraub, P.G., S. Pivonia, and A. Gera. 2008. Physical control of leafhoppers. J Econ Entomol. 101: 1337–1340.

Xu, X., Borgemeister, C. and Poehling, H.-M. 2006. Interactions in the Biological Control of Western Flower Thrips *Frankliniella occidentalis* (Pergande) and Two-Spotted Spider Mite *Tetranychus urticae* (Koch) by the Predatory Bug Orius insidiosus (Say) on Beans. Biological Control, 36, 57-64.

Yang N.W., Zang L.S., Wang S., Guo J.Y., Xu H.X., Zhang F., Wan F.H. 2014.Biological pest management by predators and parasitoids in the greenhouse vegetables in China. Biol Control 68:92–102

Evy Z Santiago
Integrated Pest Management Manual for Greenhouses
Spring 2019

Evy Z Santiago
Integrated Pest Management Manual for Greenhouses
Spring 2019