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Review of Major Crop and Animal Arthropod Pests of South Texas

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ABSTRACT

The Lower Rio Grande Valley is an area in Texas that consists of the four southern-most counties. This area contains a diverse range of agriculture and land-use including vegetable, row-crop and livestock production. The year-around cool to hot subtropical climate means that green vegetation is continually present, including many crops. Geographically, it shares an international border, making it a region vulnerable to new invasive species and the re-introduction of pests that have been previously eliminated in the United States. These combined factors lead to an array of arthropod pests that may have serious impacts on the crops, animals, and people in the region. This review focuses on arthropod pests that have historically, currently, or have the potential to significantly impact vegetables, row-crops, livestock, and humans in the LRGV. This is not an all-inclusive review but aims to focus on many of the arthropods that have been significant in the last 20 years.

Additional index words: Lower Rio Grande Valley (LRGV), insect pests, livestock pests, vegetable, row-crop

The Lower Rio Grande Valley (LRGV) is a floodplain in Southern Texas consisting of Cameron, Hidalgo, Starr and Willacy Counties. It is an incredibly diverse area with an estimated 530,300 acres in crop production, 103,500 livestock, and a human population of over 1 million (USDA, 2019). The sub-tropical and semi-arid climate allows for the successful production of a wide range of row crops, vegetables, and livestock year-around. Temperatures in the area rarely dip below 3ºC and a hard freeze is a rare occurrence (Weatherspark, 2019). This variety of plant and animal hosts combined with the lack of a prolonged cold period allow the LRGV to support a diverse range of arthropods, a few of them potentially serious pests. Another challenge in the LRGV is its shared international border or transboundary region which presents the potential threat of new invasive species as well as persistent re-introduction of, or continued pressure from, a few important pests. Thus, the LRGV remains a quarantine zone for several pests that are considered effectively eradicated in the majority of the United States. In general terms, insects and mites may feed on various plant parts including seeds, seedling plants, roots, stems, foliage, flowers, and fruits, leading to the additional time and expense of control measures as well as possible yield reduction. In addition, some of these insects may vector pathogens that cause severe economic losses in crops as well as illness and loss of life in humans and livestock.

The goal of this review is to discuss major arthropod pests of row crops, vegetables and animals. While it is not an exhaustive list, it aims to highlight selected pests that have historically, currently, or have the potential to have a serious impact on the LRGV and beyond. Each entry aims to provide a brief history of the pest, the type of damage it causes, the economic impact, and applicable integrated pest management methods.

REVIEW

Agricultural Pests

Pest
Boll weevil, A*nthonomus grandis* Boheman
Crop
Cotton
Citations: Allen, 2008; Bohmfalk et al., 1982; Metcalf et al., 1962; National Cotton Council, 2019; Smith, 1998; Texas Boll Weevil Eradication Foundation Inc., 2019; Torres and Bueno, 2018.

During the late 19th century, the boll weevil migrated into the United States from Mexico and infested all U.S. cotton-growing areas by the 1920s, devastating the industry and the people growing cotton in the American South. Adult weevils use their snout to bore into and feed on cotton terminals, buds and flowers. Females may also deposit eggs into punctures on squares and young bolls, covering the cavity with a secreted sticky substance (Bohmfalk et al., 1982; Metcalf et al. 1962.) Cotton squares and bolls may yellow and drop to the ground where larvae complete development (Metcalf et al., 1962). Larger bolls may not drop but developing lint is still damaged. The Boll Weevil Eradication Program, created in the U.S. in 1978, allowed full-scale cultivation to resume in many regions. Today, the boll weevil is considered eradicated throughout most of Texas, except in the Lower Rio Grande Valley where the Cotton Boll Weevil Eradication Foundation, Inc. continues to oversee the mandatory Boll Weevil Eradication Program (Smith, 1998). Management tactics include weekly monitoring of pheromone traps used to effectively time malathion insecticide applications, mandatory stalk destruction to prevent overwintering, and control of volunteer cotton (Allen, 2008). The extensive and repetitive use of a single insecticide creates significant pressure for the development of resistance (Torres and Bueno, 2018). It is estimated that Texas cotton producers spend approximately $70 million each year controlling the boll weevil while this pest still causes annual loses upwards of $200 million (Nat. Cotton Council, 2019). In areas where the boll weevil has been eradicated, the National Cotton Council estimates annual savings from reduced insect damage, reduced control costs, and increased yields of $80 million. Research on this important pest continues with focus on finding more effective ways to eradicate it from the Lower Rio Grande Valley.

Pest

Corn leaf aphid, *Rhopalosiphum maidis* Fitch

Crop

Corn, sorghum, barley, rye, oats, wheat, and millet

Citations: Ahman et al., 2019; Asin and Pons, 2001; Barton and Ives, 2014; Bing et al., 1991; Bitsadze et al., 2017; Faria et al., 2007; Fartek et al., 2012; Guo et al., 2017; Jasrotia et al., 2018; Kariyat et al., 2019; Louis et al., 2015; Michels and Matis, 2008; Moraes et al., 2005; Saksena et al., 1964; Sappington et al., 2018; USHDA, 1960; Vinson and Scarborough, 1991.

Corn leaf aphid (CLA) is a hemipteran insect pest that has been documented in the Rio Grande Valley as early as 1960 (USDHA, 1960). This aphid's hosts include cereal crops such as corn, sorghum and barley (Kariyat et al., 2019). They directly affect their hosts by piercing vascular organs with their needle-like mouth parts (stylet) and consuming contents (Fartek et al., 2012). For example, on corn, CLA tend to feed on the content of vascular organs located in the plant's tassel and upper leaves and thus decrease crop yield (Asin and Pons, 2001; Moraes et al., 2005). They also impact yield by vectoring various viruses including the Barley Yellow Dwarf Virus (BYVD-PAV), cucumber mosaic virus, sugarcane mosaic virus, maize dwarf mosaic virus (MDMV) and cucumber mosaic virus (Sappington et al., 2018; Guo et al., 2017). It is estimated that CLA has four major different biotypes that successfully transmit viruses (Saksena et al., 1964). They can also indirectly harm host plants by hindering photosynthesis by secreting honeydew which enables mold to grow (Jasrotia et al., 2018). *R. maidis* can also impact the ecosystem due to its mutualistic relationship with ants, including *Solenopsis invict*a, the invasive red imported fire ant (Vinson and Scarborough, 1991; Barton and Ives, 2014).

Various measures can be taken to minimize the negative impact of *R. maidis*, including biological control, pesticides and resistant crops. Natural predators such as parasitoid wasps, and coccinellid beetles have been implemented as biological controls against *R. maidis* (Faria et al., 2007; Michels and Matis, 2008). Pesticides, selective such as *Bt* toxin based or broad spectrum such as synthetic pyrethroids, may be used but these measures are mostly for managing the pest and vectored diseases rather than eradication. However, preventative measures against this pest can be taken before seedling germination and establishment, especially since substantial yield loss is experienced when the aphid infests seedlings (Bing et al., 1991). Seeds can be treated with an insecticide or mutualistic fungal species or, planting resistant crop varieties in order to diminish the impact (Bitsadze et al., 2017; Ahman et al., 2019). Advances in molecular biology and chemical ecology has found additional plant defense responses including ethylene and Jasmonic acid mediated phytohormone signaling pathways in maize (Louis et al., 2015) and resistance through secondary metabolites such as flavonoids in sorghum (Kariyat et al., 2019).

Pest

Corn leafhopper, *Dalbulus maidis* (DeLong & Wilcott)

Crop

Corn

Citations: Pitre, 1967; Nault and Bradfute, 1979; Nault and Madden, 1985.

The corn leafhopper is an insect pest found on corn from Mexico to South America. The insect species, which is believed to have originated in Mexico (Pitre, 1967), can only complete its life cycle in corn and teosintes (*Zea* spp. that are the wild progenitors of domesticated corn) making corn the only plant host of economic importance (Pitre, 1967; Nault and Madden, 1985). This insect pest causes yield losses by its direct feeding and oviposition damage and indirectly by transmitting three different pathogens: two corn-stunting mollicutes, *Maize bushy stunt phytoplasma* and the corn stunt spiroplasma, *Spiroplasma kunkelii*, in addition to the plant infecting-virus *Maize rayado fino virus* (Nault and Bradfute, 1979). This insect pest and plant pathogen vector was present in the southern United States, including the Lower Rio Grande Valley in South Texas, during the mid- to late 1990s (Badillo-Vargas, personal communication). However, the corn leafhopper seems to have been completely absent or present in low numbers in South Texas for almost two decades until its reappearance during the summer of 2016 when it was found in one commercial and one experimental corn field in Weslaco, TX (Jones, Esparza-Diaz, and Badillo-Vargas, personal communication). More recently, this insect has been observed on corn during the fall of 2018 (Wayadande and Badillo-Vargas, personal communication). However, no reports of pathogens potentially vectored by this pest were reported.

Pest

Cotton aphids: Cotton aphid, *Aphis gossypii* (Glover)
Green peach aphid, *Myzus persicae* (Sulzer)
Cowpea aphid, *Aphis crassivora* Koch

Crop

Cotton

Citations: Corrêa et al., 2005; Isakeit, 2019; Silva et al., 2008, Vyavhare and Kerns, 2017a.

While several species of aphids may feed on cotton, including green peach, cowpea and cotton aphids, they typically only become a concern when a broad-spectrum insecticide application destroys natural enemy populations that typically keep aphid populations in check (Vyavhare and Kerns, 2017a). Intense aphid pressure can stress plants, cause squares and small bolls to drop, or lead to smaller bolls with incomplete fiber development (Vyavhare and Kerns, 2017a). *Aphis gossypii* is capable of vectoring cotton leaf roll dwarf virus, also known as cotton blue disease (Corrêa et al., 2005). This disease, which is only known to be transmitted by aphids, was originally discovered in the Central African Republic in 1949 (Silva et al., 2008). It was first detected in the United States in 2017 in Alabama and has since been found in Florida, Georgia, Mississippi, and South Carolina (Isakeit, 2019). At the time of publication, it is not an imminent threat to Texas cotton as it has not been detected in any commercial cotton fields, but growers should be on the lookout for symptomatic plants.

Pest

Diamondback moth, *Plutella xylostella* (L.)

Crop

Crucifers

Citations: Capinera, 2001; Cartwright et al., 1987; Harcourt, 1963; Philips et al., 2014; Shelton et al., 2000.

The diamondback moth originated in South Africa and the Mediterranean but is now a cosmopolitan pest of all cruciferous growing regions of the world (Harcourt, 1963). Thus, larvae are potential pests of all cruciferous crops in the LRGV. Because larvae are small and feed on the underside of leaves, damage is often not noted until larval development is nearly complete. Large populations can cause significant defoliation and may disrupt head formation in cabbage, broccoli, and cauliflower (Philips et al., 2014). Even minor feeding damage or the presence of larvae on market produce can lead to total rejection (Capinera, 2001). Effective control of diamondback moth larvae using insecticides often requires multiple sprays and they have developed some level of resistance to almost every class of insecticide (Shelton et al., 2000), thus, it is very important to rotate between different modes of action (Cartwright et al., 1987).

Pest

False chinch bug, *Nysius raphanus* Howard

Crop

Crucifers, cotton, sorghum

Citations: Cranshaw, 2013; Demirel and Cranshaw, 2006; Leigh, 1961; Wood and Starks, 1972.

False chinch bugs prefer canola, mustards, cabbage, and other cruciferous plants but are highly polyphagous and therefore are a potential pest of several South Texas crops. Populations may build up in sesame, canola and other mustard crops then migrate in huge numbers into adjacent cotton, onion, and sorghum fields (Cranshaw, 2013). They feed with piercing-sucking mouthparts and may overwhelm seedling plants, causing them to wilt and even die (Leigh, 1961). Damage is probably most common in canola where feeding on flowers and seedpods later in the season can cause significant injury (Demirel and Cranshaw, 2006). Feeding directly on heads in sorghum, while uncommon, can have detrimental effects on yield (Wood and Starks, 1972).

Pest

Potato psyllid, *Bactericera cockerelli* (Sulc)

Crop

Chipping potatoes and other Solanaceae

Citations: Goolsby et al., 2007; Goolsby et al., 2012 ; Guenther et al,. 2012 ; Munyaneza et al., 2007; Workneh et al. 2018.

The potato psyllid vectors the zebra chip disease and is a major pest of chipping potatoes in the Rio Grande Valley (Guenther et al. 2012). It was first documented in 2007 and an IPM program was developed to control the pest to prevent disease transmission (Munyaneza et al., 2007; Goolsby et al., 2007, Goolsby et al., 2012). Approximately 3,000 acres of potatoes are impacted in Hidalgo Co. (Guenther et al., 2012). Potato psyllid is also a minor pest on other solanacecous crops such as tomato, tomatillo, and pepper, which when infected by the disease-causing agent, display a disease commonly known as psyllid yellows (Workneh et al., 2018).

Pest

Spider mites:
Two-spotted spider mite, *Tetranychus urticae* Koch
Banks grass mite, *Oligonychus pratensis* (Banks)

Crop

Cotton, corn, soybean, vegetables and others

Citations: Cronholm et al., 1998; Drees and Jackman, 1998; Hinomoto et al. 2001; Vyavhare et al., 2018.

Spider mites are prevalent pests in cotton, corn, soybeans, citrus, vegetables and many other crops in the Valley. There are two well-known species of spider mites widely seen in most crops in the LRGV: Banks grass mite and the two-spotted spider mite. There is a third mite, commonly referred to as the carmine spider mite that is now recognized as a red form of the two-spotted spider mite (Hinomoto et al. 2001).

Both color forms of the two-spotted mite are most common in cotton whereas Banks grass mite is known for feeding primarily in corn and sorghum (Vyavhare et al., 2018). Adult mites are small in size (0.4mm), making detection difficult. Their life cycle from egg to adult generally takes 5 to 20 days depending on the temperature and females can lay about 50 clear eggs resulting in rapid population growth (Drees and Jackman, 1998). Both nymphs and adults pierce plant tissues with their stylets, extracting juices (chloroplasts) out of the plant cells, leading to cell death (Vyavhare et al., 2018). They feed on the undersides of leaves along the midrib. Early damage can be detected on the top of leaves and will appear as yellow or red spotting. Continued infestations result in bronzing of the entire leaf and eventually, defoliation. As their name suggests, heavy infestations of spider mites result in a fine webbing on the underside of the leaves. Spider mite pressure can occur at any time during the season, but they tend to be favored by hot, dry weather. Infestations tend to be sporadic in fields but when heavy infestations of spider mites are present, they can cause stunted plant growth and significant yield loss (Cronholm et al., 1998).

Pest

Sugarcane aphid, *Melanaphis sacchari* (Zehntner)

Crop

Grain and forage sorghum, sorghum/sudan hybrids, sugarcane

Citations: Armstrong et al., 2015; Bowling et al., 2016; Kaur et al., 2020; Kerns et al., 2015; Knutson et al., 2016; Michaud et al., 2016; Wilson, 2019.

The sugarcane aphid (SCA) is an invasive hemipteran insect that originated from Africa and the Middle East but became a significant problem for TX sorghum growers in 2013 (Bowling et al., 2016). Like other aphids, SCA can reproduce asexually, giving birth to live clones, thus populations grow very quickly. These aphids damage sorghum by removing plant sap from the underside of leaves and producing copious amounts of honeydew leading to sooty mold buildup (Knutson et al., 2016). Significant infestations may lead to plant death, reduced numbers of heads, reduced seed weight, delayed development, and reduced harvest efficiency due to excessive honeydew (Bowling et al., 2016; Kerns et al., 2015). Two products, sulfoxaflor and flupyradifurone have been found to provide high aphid mortality with a minimal impact on beneficial arthropods, mitigating secondary pest outbreaks and SCA resurgence (Michaud et al., 2016). Commercially available sorghum hybrids with some level of aphid resistance are available with continued breeding for host plant resistance underway (Bowling et al., 2016). SCA can occur on both crop and non-crop plants (e.g. sorghum and sugarcane) as well as Johnsongrass. In addition to this damage, these aphids have been identified as vectors of the Sugarcane Yellow Leaf Disease (SCYL) (Knutson et al., 2016). This is a significant threat to agriculture in the U.S. and Mexico which produce over 1.1. million ha. of sugarcane (Wilson, 2019). Crop loss from their foliar feeding and disease transmission, has made the SCA an economically important insect, especially in sorghum in the United States. (Armstrong et al., 2015; Kaur et al., 2020).

Pest

Stinkbugs, multiple species

Crop

Cotton, soybeans

Citations: Hopkins et al., 2009; Sosa-Gómez et al., 2019; Vyavhare et al., 2014; Wene and Sheets, 1964; Whitworth and Davis, 2009.

Stink bugs use piercing-sucking mouthparts to suck sap from succulent tissue in a wide variety of plants and can become an economic pest when they feed directly on pods in soybeans and bolls in cotton (Vyavhare et al., 2014, Whitworth and Davis, 2009). Studies indicate that the most common species in the region are brown stink bugs including *Euschistus servus* (Say), *E. Quadrator* Rolston and *E. obscurus (Palisot*), along with the southern green stink bug, *Nezara viridula* (L.), and green stink bug, *Chinavia hilaris* (Say) (Hopkins et al., 2009). In addition, an invasive species, the redbanded stink bug, *Piezodorus guildinii* (Westwood) has become a potential threat to soybeans and cotton due to its wide host range and good mobility (Vyavhare et al., 2014). In cotton, stink bugs pierce bolls and feed, causing small bolls to become soft and yellow leading to damaged and shriveled seeds and stained lint (Wene and Sheets, 1964). In soybeans, feeding on developing seeds within pods can lead to small, shriveled and deformed seeds (Whitworth and Davis, 2009). Feeding punctures expose bolls and pods to moisture, air, and pathogens (Whitworth and Davis, 2009). Stink bugs can be difficult to manage with foliar insecticide applications and many failures have been reported, in many cases due to the development of resistance (Sosa-Gómez et al., 2019). Increased resistance to commonly used organophosphates, neonicotinoids, and pyrethroids has been documented (Sosa-Gómez et al., 2019) Annual losses due to stink bugs in south Texas have ranged from just over $160,000 to more than $4 million (Hopkins et al., 2009).

Pest

Texas potato beetle, *Leptinotarsa texana* Schaeffer-

Crop

Solanaceous crops

Citations: Cuda et al., 2002; EPPO Bulletin, 2007; Hsiao, 1988; Kariyat and Chavana, 2018; Lefoe et al., 2020; Olckers et al., 1995 and 1999; Pallister, 1953; Petanidou et al., 2018; Sperling and Mitchell, 1991.

The Texas potato beetle is commonly found in South Texas (including the Lower Rio Grande Valley) and has a range that extends from Missouri through Texas into Mexico (Pallister, 1953). Beetles of the genus *Leptinotarsa* are specialist feeders of host plants that belong to the Solanaceae family (Hsiao, 1988). The beetle feeds on silver leaf nightshade, *Solanum eleaegnifolium*, a species native to the Rio Grande

Valley, but highly invasive in other parts of the world (EPPO Bulletin, 2007; Petanidou et al., 2018). Both the adults and the grubs feed on all parts of *S. eleaegnifolium* and have shown some host specificity towards this species, and a few other members of the family (Kariyat and Chavana, 2018). Thus, *L. texana* has been implemented as a biocontrol agent for silver leaf nightshade in South Africa (Sperling and Mitchell, 1991; Olckers et al., 1995 and 1999) and is currently tested in Australia (Lefoe et al., 2020). It has economic importance in the United States, as well as in other parts of the world due to its voracious feeding, primarily on the noxious weed, *S. eleaegnifolium* as well as crops that belong to the Solanaceae family including tomato, potato and eggplant (Cuda et al., 2002).

Pest

Thrips: Onion thrips, *Thrips tabaci* Lindeman
Western flower thrips, *Frankliniella occidentalis* (Pergande)
Bean thrips, *Caliothrips fasciatus* (Pergande)

Crop

Cabbage, cotton, onion

Citations: Diaz-Montano et al., 2011; Greenberg et al., 2009; Riley et al., 2011; Vyavhare and Kerns, 2017b; Riley et al., 2011.

There are several species of economically important thrips including onion thrips, western flower thrips, and bean thrips (Greenberg et al., 2009). Presence of a particular species depends largely on the crop being grown, or crops in the near vicinity. Onion thrips are a significant pest of onions where feeding on leaves causes blotches, premature senescence and distorted, undersized bulbs resulting in up to 50% yield loss (Diaz-Montano et al., 2011). Thrips often migrate to germinating cotton fields after onion harvest where they feed on leaves, leaf buds, and small squares. Damage includes deformed leaves and destroyed terminal buds creating excessive branching, and delayed growth (Vyavhare and Kerns, 2017b). Thrips may make cabbage unmarketable due to the blistered, bronzed, and scarred appearance caused by their feeding. Additionally, thrips are capable of vectoring tospoviruses including tomato spotted wilt virus and iris yellow spot virus, to various crops, potentially leading to devastating yield losses (Riley et al., 2011). For example, in the U.S., tomato spotted wilt virus caused and estimated $1.4 billion in losses over a 10-year period (Riley et al. 2011). An outbreak of iris yellow spot virus led to 60-70% onion yield losses in the mid-2000's (Anciso, personal communication).

Pest

Tomato bug, *Nesidiocoris tenuis* (Reuter)

Crop

Sesame and tomato

Citations: Albajes et al., 2006; Calvo et al., 2009; Hoberlandt, 1955; Sanchez et al., 2014; Urbaneja et al., 2009.

The tomato bug, of Mediterranean origin, is an invasive pest of sesame and tomato (Hoberlandt, 1955). It has a zoophytophagous feeding behavior that allows it to feed on plants (=phyto) as well as on other small arthropods (=zoo) (Calvo et al., 2009; Sanchez et al., 2014). This voracious general predator is frequently used to control several insect pests in greenhouses such as the silverleaf/sweetpotato whitefly and the greenhouse whitefly (Albajes et al., 2006). Nonetheless, this insect could be a devasting pest in crops such as sesame and tomato (Urbaneja et al., 2009). The tomato bug was initially detected in South Texas in 2012 (Esparza-Diaz and Villanueva, personal communication), and its presence in subsequent years (2013 through 2019) clearly shows that this exotic species is now established in the Lower Rio Grande Valley in South Texas. Under certain circumstances, however, this insect can be a biological control agent of agriculturally important insect pests and vectors as is the case for potato psyllids in potatoes (Esparza-Diaz and Badillo-Vargas, personal communication).

Pest

Whiteflies: Silverleaf whitefly, *Bemisia argentifolii* Bellows and Perring or *Bemisia tabaci* biotype B
Bandedwing whitefly, *Trialeurodes abutilonea* (Haldeman)

Crop

Cotton, crucifers, cucurbits, ornamental plants, tomatoes, other Solanaceae

Citations: Bellows et al., 1994; Bi et al., 2001; Ciomperlik and Goolsby, 2017; Goolsby et al., 1998; Goolsby et al., 2005; Henneberry and Faust, 2008; Hogenhout et al., 2008; Horowitz et al., 2020; Jones, 2003; Zhang et al., 2011.

Silverleaf whitefly and Bandedwing whitefly attack a broad range of host plants. The silverleaf or sweet potato whitefly, *Bemisia tabaci* Biotype B, is specifically the only biotype present thus far in the Lower Rio Grande Valley in Texas (Bernal Jimenez and Badillo-Vargas, personal communication) and is considered one of the most important cotton pests worldwide (Zhang et al., 2011). The silverleaf/sweet potato whitefly has a significant economic impact on United States' agriculture, approaching $10 billion from 1980 to 2000, due to crop loss, job displacement, and cost of control (Henneberry and Faust, 2008). In cotton, feeding by these sucking insects may lead to stunted plants, cotton defoliation, boll shed, and reduced yields (Bellows et al., 1994; Bi et al., 2001). If honeydew is deposited on fiber it creates 'sticky' cotton which causes problems during lint processing (Bellows et al., 1994; Bi et al., 2001). When cotton is defoliated large whitefly populations may move into various cruciferous crops where they may cause stem blanching and whitening (Henneberry and Faust, 2008). Additionally, issues related to the growth of sooty mold on secreted honeydew can interfere with photosynthesis. Whiteflies are known to vector over 100 plant viruses worldwide, including Tomato yellow leaf curl virus, which severely limits tomato production in South Texas (Jones, 2003; Hogenhout et al., 2008). Insecticide resistance is an ongoing problem in the control of whiteflies as resistance has been detected to more than 60 active ingredients and most major classes of insecticides (Horowitz et al., 2020). Biological control has played an important role in mitigating whitefly problems in the LRGV. The USDA implemented a national program including the release of whitefly parasitoids in the mid-1990's that resulted in the successful establishment of *Eretmocerus hayati* (Goolsby et al., 1998; Ciomperlik and Goolsby, 2017). This success is likely due to the parasitic wasp's narrow host range and high attack rate as well as the climatic match of the LRGV to its native Pakistan (Goolsby et al., 2005). Good IPM practices have allowed this parasitoid to provide consistent control of whitefly populations. However, cotton defoliation and broad-spectrum insecticide applications can suppress parasitoids which leads to occasional whitefly outbreaks (Ciomperlik and Goolsby, 2017).

Livestock and Medical Pests

Pest
American Dog Tick, *Dermacentor variabilis* (Say)
Host
Dog, Coyote, Horses, Cattle, Human
Citations: Dantas-Torres, 2007; Kocan et al. 1992, Sonenshine, 2018; Stiller and Coan, 1995.

The American dog tick is the most important vector of the causative agent of Rocky Mountain Spotted Fever, *Rickettsia ricketsii* in the United States (Sonenshine, 2018). It is the deadliest tick-borne disease in the US primarily because it is difficult to diagnose in the early stages (Dantas-Torres, 2007) and is often lethal if untreated. This is a 3-host tick and although called the dog tick, it has a wide host range. Canines are its preferred host but they will also bite livestock, pets and humans. It also vectors the agents of Equine babesiosis (*Babesia caballi*), Equine piroplasmosis (*Theileria equi*) and Bovine Anaplasmosis (*Anaplasma marginale*) (Stiller and Coan, 1995). Interestingly, the latter disease is vectored almost exclusively by the male ticks (Kocan et al. 1992).

Pest
Black-Legged Tick, *Ixodes scapularis* Say
Host
Humans, White-tailed deer
Citations: Billings et al., 1998; Burgess and Lindberg, 1989; Dahlgren et al., 2011; Dennis et al., 1998; Feria-Arroyo et al., 2014; Mitchell et al., 2016; Pepin et al., 2012; Syzoni et al., 2015; Waldrup et al., 1992. Williamson et al., 2010; Zhang et al., 2006.

The black-legged tick is a 3-host tick with deer being the typical host of the adult stage. It is the primary vector of Lyme disease over the northeastern and midwestern U.S. The CDC estimates the economic impact of Lyme Disease in the U.S. at $203 million annually (Zhang et al. 2006). The incidence and distribution of the clinical disease in Texas suggests that many cases were acquired elsewhere or are misdiagnoses (Szyoni et al., 2015). The distribution map by Dennis et al. (1998) does not show *Ixodes scapularis* extending into south Texas, however Feria-Arroyo et al. (2014) confirm that the tick and the Lyme disease agent is present on deer in south Texas and northern Mexico. The larval hosts for this tick are usually rodents, but it prefers reptiles in the southerly part of its range (Pepin et al., 2012). The larval host in the Rio Grande Valley is unknown; surveys here have only found the adults. The difference is critical as amplification of the etiological agent occurs in small mammals. Surveys have shown that a large percentage of coyotes and rabbits are seropositive for the Lyme disease agent in Texas (Burgess and Lindberg, 1989). This tick species is also the vector of human granulocytic anaplasmosis (Dahlgren et al., 2011), human Ehrlichiosis and human Babesiosis (Mitchell et al., 2016; Williamson et al., 2010). Regarding wildlife, *I. scapularis* has been implicated as the vector of *Babesia odocoilei* in Texas (Waldrup et al., 1992) and uniquely in Texas it is the vector of *Rickettsia cooleyi*, a non-pathogenic organism (Billings et al., 1998), but whose presence complicates diagnostic testing.

Pest
Cattle fever ticks, *Rhipicephalus (Boophilus)* sp.
Host
Cattle, nilgai, white-tailed deer, horses
Citations: Esteve-Gassent, et al., 2014; Foley et al., 2017; Goolsby et al., 2016; Graham and Hourrigan, 1977; Pérez de León et al., 2012; Singh et al., 2018, Tidwell et al. 2019; USDA, Texas Cattle Fever.

Cattle fever ticks, *Rhipicephalus* (=*Boophilus*) *microplus* Canestrini and *Rhipicephalus—(Boophilus) annulatus* (Say) have a wide geographic distribution, spanning tropical and subtropical regions between parallels 32°N latitude and 35°S latitude (Goolsby et al., 2016). These livestock pests cause huge economic losses to milk and meat production, both directly (reduced weight gain and milk production, anaemia, and, mortality) and indirectly by inducing quarantines and transmission of various disease-causing pathogens such as *Babesia* spp. and *Anaplasma* spp. (Pérez de León et al., 2012). Cattle fever ticks and bovine babesiosis are estimated to have caused losses to the U.S. livestock industry close to $3 billion annually in today's currency before they were eradicated from the U.S. (Graham and Hourrigan, 1977; USDA, Texas Cattle Fever). An eradication program based on continuous surveillance by mounted inspectors and the use of acaricides has been implemented in the U.S. along the Texas-Mexico border to manage periodic outbreaks (Tidwell et al. 2019). However, novel strategies are needed for the continued detection, suppression and eradication of cattle fever ticks in the permanent quarantine zone due to the emerging role of 1) exotic nilgai antelope, *Boselaphus tragocamelus* (Pallas) and white-tailed deer, *Odocoileus virginianus* (Zimmerman) as cattle fever tick hosts; 2) growing evidence of acaricide resistance; and 3) the invasion of pathogenic landscape-forming species such as the giant reed, *Arundo donax*, and other exotic plant species that contribute to suitable habitat for cattle fever ticks

(Esteve-Gassent et al., 2016; Foley et al., 2017; Pérez de León et al., 2012; Singh et al. 2018).

Pest

Cayenne Tick, *Amblyomma mixtum* Koch

Host

Cattle, Deer

Citations: Mitchell et al., 2016; Nava et al., 2014; Scoles et al., 2011; Williamson et al., 2010

Technically a misnomer because the true cayenne tick, *Amblyomma cajennense* (F.), is restricted to parts of South America (the Guianas, *fide* Nava et al., 2014). The species found in the Rio Grande Valley, *Amblyomma mixtum* Koch occurs from south Texas to Ecuador. A 3-host tick, adults have been recorded on cattle, horses, donkeys, pigs, dogs and humans (Strickland et al., 1976). Wild hosts include coyote, squirrel, armadillo, tapir, peccary and badger (Guzman -Cornejo et al., 2011). However, it is most abundant on white-tailed deer. As *A. cajennense* in Texas it has been implicated as the vector of equine piroplasmosis (Scoles et al., 2011), human Ehrlichiosis, and *Borrelia lonestari*, the suspected agent of southern tick associated rash illness (Mitchell et al., 2016; Williamson et al., 2010).

Pest

Gulf-Coast Tick, *Amblyomma maculatum* Koch

Host

Cattle, Horses, Sheep, Goats

Citations: Cooley and Kohls 1944; Lada et al., 2018; Mitchell et al., 2016; Parker et al., 1939; Teel et al., 2010.

The gulf coast tick is widespread, occurring across the southern US from Florida to California south to Paraguay (Lada et al., 2018). They are an important pest of cattle infesting sheep and horses as well (Cooley and Kohls, 1944) and is another tick that prefers the ears as the bite site. A 3-host tick, the larvae infest primarily birds. They harbor *Rickettsia* in the spotted fever group (Parker et al., 1939) and human Ehrlichiosis (Mitchell et al., 2016). Experimentally it is an efficient vector of heartwater fever, a disease introduced into the Caribbean region on African cattle, thus posing an existential threat to the livestock industry on the American mainland (Teel et al., 2010). The latter estimate annual losses of $58 million due to infestations of this tick on livestock.

Pest

Tropical Horse Tick, *Anocentor nitens* (Neumann)

Host

Horses, Donkeys, White-tailed Deer

Citations: Barbosa et al., 1995; Borges, 2000; Hooker et al., 1912; Schwint et al., 2008; Stiller and Coan, 2008.

The tropical horse tick is the most important ectoparasite of horses and donkeys in Latin America (Borges, 2000). It is the vector of equine babesiosis and equine piroplasmosis (Barbosa et al., 1995; Schwint et al., 2008). There is also evidence that they are at least potential vectors of bovine Anaplasmosis (Stiller and Coan, 1995) although they are rarely found on cattle. The distribution *A. nitens* is south Texas to Argentina and the Caribbean Islands where it was probably introduced. It became established in Florida in 1958. It is a one-host tick and aside from its usual equine host has also been reported on cattle and dogs. The only sylvatic host record is a solitary report on a deer hide (Hooker et al., 1912). In fact, they heavily infest deer at the Laguna Atascosa National Wildlife Refuge in Cameron county Texas. Inasmuch as horses, cattle and dogs are not native to the New World. The common occurrence of this tick on white-tailed deer on the coastal areas of south Texas suggests that deer are the likely native host. On both horses and deer, the preferred infestation site is in the ears.

Pest

Mosquitoes

Yellow fever mosquito, *Aedes aeypti* (Linneaus)

Asian tiger mosquito, *Aedes albopictus* (Skuse)

Southern house mosquito, *Culex quinquefasciatus* Say

Host

Humans and other animals

Citations: Champion and Vitek, 2014; Dye-Braumuller et al., 2019; Martin et al., 2019; Sames et al., 1996

Mosquitoes, including *Aedes aegypti*, *Ae. albopictus*, and *Culex quinquefasciatus* are present in the Lower Rio Grande Valley in South Texas indoors and outdoors all year long; although their abundance and distribution fluctuate with temperature, precipitation, and land use patterns (Champion and Vitek, 2014). These three species of mosquitoes are known vectors of arboviruses and, along with other mosquito species, act as nuisance pests. The species *Aedes aegypti* and *Ae. albopictus* both are known to transmit Dengue virus, Chikungunya virus, and Zika virus with humans acting as the reservoir host, while *C. quinquefasciatus* transmits West Nile virus, St. Louis encephalitis, and Western equine encephalitis (Martin et al., 2019), which have zoonotic hosts including local bird species. Either in response to nuisance pests calls or due to their disease transmission surveillance, mosquito control is usually observed at dawn or dusk in different areas in the Lower Rio Grande Valley, often targeting *Culex* species of mosquitoes. Vector control may consist of larviciding and adulticiding (*i.e.,* the process to control the immature and mature mosquito stage, respectively) in critical in areas that have been deemed "hot spots" for arbovirus introduction and establishment, such as the Mexico – Texas border, to protect human health, although other control methods such as habitat reduction work well for *Ae. aegypti* and *Ae. albopictus*. Adulticiding often consists of utilizing backpack sprayers or truck-mounted ULV sprays to target resting adult mosquitoes, while larviciding will introduce pesticides to the aquatic larval habitat (often consisting of granules, pellets, or "donut-like" dunks) (Dye-Braumuller et al., 2019). Concerns about the evolution of resistance to commonly used insecticides (Sames et al. 1996) necessitate the implementation of new strategies and control efforts. Recent efforts to expand control using integrated pest management (IPM) strategies such as removing breeding or resting habitat as well as limiting interaction between humans and mosquitoes through changes in human behavior or use of repellents (Dye-Braumuller et al., 2019).

DISCUSSION

The pests discussed in this review represent a wide range of arthropods with the potential to impact the crops, animals, and people of the LRGV. Some of these pests are present in economically important numbers every year, while others may be present at low levels until favorable conditions allow populations to reach levels that require control. Some of these arthropods do not typically directly impact crops but may vector diseases that lead to significant yield losses. Regardless, all the arthropod pests in this review have historically required, and will continue to require, the development of integrated pest management methods to mitigate their damage. These methods include breeding for host plant resistance, the development of new, more selective insecticides to combat insecticide resistance, cultural controls such as planting time and destruction of volunteer plants, the development of effective biological control programs, as well as continued quarantine and eradication programs for particular pests.

LITERATURE CITED

Åhman, I., S. -Y. Kim, and L. -H. Zhu. 2019. Plant genes benefitting aphids—potential for exploitation in resistance breeding. Front. in Plant Sci. 10. doi: 10.3389/fpls.2019.01452

Albajes, R., C. Castañé, R. Gabarra, and O. Alomar. 2006. Risk of plant damage caused by natural enemies introduced for arthropod biological control. Pp. 132-144 in Bigler, F., D. Babendreier, and U. Kuhlmann. (Eds) *Environmental Impact of Invertebrates for Biological Control of Arthropods: Methods and Risk Assessment*. Oxon, UK, CABI Publishing.

Allen, C. T. "Boll weevil eradication: an areawide pest management effort". *Areawide Pest Management Theory and Implementation.* Ed. Koul, O., G. W. Cuperus, and N. Elliott. Cambridge, MA: CABI, 2008. Pgs. 467 – 559.

Armstrong, J. S., W. L. Rooney, G. C. Peterson, T. Villenueva, M. J. Brewer and D. Sekula-Ortiz, D. 2015. Sugarcane aphid (Hemiptera: Aphididae): host range and sorghum resistance including cross-resistance from greenbug sources. J. Econ. Entomol. 108: 576-582.

Asin, L., and X. Pons. 2001. Effect of high temperature on the growth and reproduction of corn aphids (Homoptera: Aphididae) and implications for their population dynamics on the Northeastern Iberian Peninsula. Environ. Entomol. 30: 1127-1134.

Barbosa, P., B. R. Peymann, and K. T. Friedhoff. 1995. Epidemiological aspects of equine babesiosis in a herd of horses in Brazil. Vet. Parasitol. 58: 1-8.

Barton, B. T., and A. R. Ives. 2014. Direct and indirect effects of warming on aphids, their predators, and ant mutualists. Ecology. 95: 1479–1484. doi: 10.1890/13-1977.1

Bellows, Jr. T. S., T. M. Perring, R. J. Gill, and D. H. Headrick. 1994. Description of a species of *Bemisia tabaci* (Homoptera: Aleyrodidae). Annu. Entomol. Soc. of Am. 78: 195-260.

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

Billings, A.N., G.J. Teltow, S.C. Weaver, and D.H. Walker. 1998. Molecular characterization of a novel *Rickettsia* species from *Ixodes scapularis* in Texas. Emerg. Infect. Dis. 4: 305-309.

Bing, J. W., M. G Novak, J. J. Obrycki, and W. D. Guthrie. 1991. Stylet penetration and feeding sites of *Rhopalosiphum maidis* (Homoptera: Aphididae) on two growth stages of Maize. Ann. Entomol. Soc. Amer., *84*: 549–554. doi: 10.1093/aesa/84.5.549

Bitsadze, N., K. Pavliashvil, R. Khazaradze, M. Beruashvili, and M. Betsiashvili. 2017. Preliminary study if selected entomopathogenic fungi for corn leaf aphid, *Rhopalosiphum maidis*, control. Microb.l and Nemat. Control of Invert. Pests. *129*: 38–42.

Bohmfalk, G. T., R. E. Frisbie, W. L. Sterling, R. B. Metzer, and A. E. Knutson. 1982. Identification, biology, and sampling of cotton insects. Texas Agricultural Extension Service, Texas A&M University System.

Borges, L. M. F., P. R. Oliveira, and M. F. B. Ribeiro. 2000. Seasonal dynamics of *Anocentor nitens* on horses in Brazil. Vet. Parasitol. 89: 165-171.

Bowling, R. D., M. J. Brewer, D. L. Kerns, J. Gordy, N. Seiter, N. E. Elliot, G. D. Buntin, M. O. Way, T. A. Royer, S. Biles, and E. Maxson. 2016. Sugarcane aphid (Hemiptera: Aphididae): a new pest on sorghum in North America. J. Integr. Pest Manag. 7: 12: 1-13.

Burgess, E.C. and L.A. Lindberg. 1989. Borrellia sp. infection in coyotes, black-tailed jack rabbits and desert cottontails in southern Texas. J. Wildlife Dis. 25: 47-51.

Calvo, J., K. Bolckmans, P. A. Stansly, and A. Urbaneja. 2009. Predation by *Nesidiocoris tenuis* on *Bemisia tabaci* and injury to tomato. BioControl. 54: 237-246.

Capinera, J. L. 2001. Handbook of Vegetable Pests. Academic, New York, NY, p. 729.

Cartwright B., J. V. Edelson, and C. Chambers. 1987. Composite action thresholds for the control of lepidopterous pests on fresh-market cabbage in the lower Rio Grande Valley of Texas. J. Econ. Entomol. 80: 175-181.

Cartwright, B., J. V. Edelson, and C. Chambers. 1987. Composite action thresholds for the control of lepidopterous pests on fresh-market cabbage in the Lower Rio Grande Valley of Texas. J. Econ. Entomol., 80, 175-181.

Champion S. and C.J. Vitek. 2014. Aedes aegypti and Aedes albopictus habitat preferences in South Texas. Environ. Health Insights. 8 (Suppl 2): 35-42.

Ciomperlik, M. and J. Goolsby. 2017. Whiteflies in South Texas agriculture, a historical perspective. The AgMag. Sept./Oct. 2017: pgs 22-23. Print.

Cooley, R. A., and G. M. Kohls. 1944. The genus *Amblyomma* in the United States. J. Parasitol. 30: 77-111.

Corrêa, R. L., T. F. Silva, J. L. Simoes-Araujo, P. A. V. Barroso, M. S. Vidal, and M. F. S. Vaslin. 2005. Molecular characterization of a virus from the family Luteoviridae associated with cotton blue disease. Arch. Virol. 150: 1357 – 1367.

Cranshaw, W. 2013. False Chinch Bugs. Colorado State University Extension. Fact Sheet 5.603.

Cronholm, G., A. Knutson, R. Parker, G. Teetes, and B. Pendleton. 1998. Managing Insect and Mite Pests of Texas Sorghum. Texas Agric. Ext. Ser. Bul. B-1220.

Cuda, J. P., P. E. Parker, B. R. Coon, F. E. Vasquez, and J. M. Harrison. 2002. Evaluation of exotic *Solanum* spp. (Solanales: Solanaceae) in Florida as host plants for the leaf beetles *Leptinotarsa defecta* and *L.* texana (Coleoptera: Chrysomelidae). Fla. Entomol. 85: 599-610.

Dahlgren F. S., E. J. Mandel, J. W. Krebs, R. F., Massung and J. H. McQuiston. 2011. Increasing incidence of *Ehrlichia chaffeensis* and *Anaplasma phagocytophilum* in the United States, 2000-2007. Am. J. Trop. Med. Hyg. 85: 124-131.

Dantas-Torres, F. 2007. Rocky Mountain Spotted Fever. Lancet Infect. Dis. 7:724-732.

Demirel, N., and W. Cranshaw. 2006. Plant yield response to artificial infestation of the false chinch bug, *Nysius raphanus*, confined on spring canola. Phytoparasitica. 34: 477-485.

Dennis, D. T., T. S. Nekomoto, J. C. Victor, W. S. Paul and J. Piesman. 1998. Reported distribution of *Ixodes scapularis* and *Ixodes pacificus* (Acari: Ixodidae) in the United States. J. Med. Entomol. 35: 629-638.

Diaz-Montano, J., M. Fuchs, B. A. Nault, J. Fail, A. M. Shelton, and M. Anthony. 2011. Onion thrips (Thysanoptera: Thripidae): A global pest of increasing concern in onion. J. Econ. Entomol. 104: 1-13.

Drees, B. and J. Jackman. 1998. A field guide to common Texas insects – Spider Mite. 28 January 2020. <https://texasinsects.tamu.edu/spider-mite/>

Dye-Braumuller, K., C. Fredregill, and M. Debboun. 2019. Mosquito Control. Pp 250-279 in M. Debboun, M.R. Nava, and L.M. Rueda (eds) *Mosquitoes, Communities, and Public Health in Texas*. London, UK, Academic Press (Elsevier).

EPPO Bulletin. *Solanum elaeagnifolium*. 2007. EPPO Bulletin, 37(2): 236-245. doi:10.1111/j.1365-2338.2007.01112.x

Esteve-Gassent, M.D., A. A. Perez de Leon, D. Romero-Salas, T. P. Feria-Arroyo, R. Patino, I. Castro-Arellano, G. Gordillo-Perez, A. Auclair, J. Goolsby, R. I. Rodriguez-Vivas, and J. G. Estrada-Franco. 2014. Pathogenic landscape of transboundary zoonotic diseases in the Mexico-US border along the Rio Grande. Front. Public Health. 2: 1-23.

Faria, C. A., F. L. Wäckers, J. Pritchard, D. A. Barrett, and T. C. Turlings. 2007. High susceptibility of Bt maize to aphids enhances the performance of parasitoids of lepidopteran pests. *PLoS ONE*, *2* (7), 1–11. doi: 10.1371/journal.pone.0000600

Fartek, B., S. Nibouche, P. Turpin, L. Costet, and B. Reynaud. 2012. Resistance to *Melanaphis sacchari* in the sugarcane cultivar R 365. Entomologia Experimentalis Et Applicata, 144: 270–278. doi: 10.1111/j.1570-7458.2012.01286.x

Feria-Arroyo, T. P, I. Castro-Arellano, G. Gordillo-Perez, A. L. Cavazos, M. Vargas-Sandoval, A. Grover, J. Torres, R. F. Medina, A. A. Perez de Leon, and M. D. Esteve-Gassent. 2014. Implications of climate change on the distribution of the tick vector *Ixodes scapularis* and risk for Lyme disease in the Texas-Mexico transboundary region. Parasite. Vector. 7: 199-215.

Foley, A.M., J. A. Goolsby, A. Ortega-S. Jr. J.Ortega-S. J. Sr., A. Pérez de León, N. K. Singh, A. Schwartz, D. Ellis, D. G. Hewitt, and T. A. Campbell. 2017. Movement patterns of nilgai antelope in South Texas: implications for cattle fever tick management. Prev. Vet. Med. 146: 166-172.

Goolsby, J. A., J. A. Adamczyk, Jr., B. E. Bextine, D. Lin, J. E. Munyaneza, and G. Bester, 2007. Development of an IPM program for management of the potato psyllid to reduce incidence of zebra chip disorder in potatoes. Subtrop. Plant Sci. 59: 85-94.

Goolsby, J.A., J. A. Adamczyk, J. J. Crosslin, J. M. Troxclair, N. N. Anciso, J. R. Bester, G. G. Bradshaw, J. D. Bynum, E. D. Carpio, L.A. Henne, D. C. Joshi, A. Munyaneza, J. E. Porter, P. Sloderbeck, P. E. Supak, J. R. Rush, C. M. Willett, F. J. Zechmann, B. J. and Zens, B. A. 2012. Seasonal population dynamics of the potato psyllid, *Bactericera cockerelli* (Hemiptera: Triozidae) and its associated pathogen "Candidatus Liberibacter solanacearum" in potatoes in the southern Great Plains of North America. J. Econ. Entomol. 105: 1268-1276.

Goolsby, J.A., F. D. Guerrero, J. Gaskin, K. G. Bendele, P. Azhahianambi, D. Amalin, M. Flores-Cruz, J. Kashefi, L. Smith, A. Racelis, R. K. Saini, and A. Perez de Leon. 2016. Molecular comparison of cattle fever ticks from native and introduced ranges with insights into optimal search areas for classical biological control agents. Southwest. Entomol. 41: 595-604.

Goolsby, J.A., M. A. Ciomperlik, B. C. Legaspi Jr., J. C. Legaspi, and L. E. Wendel. 1998. Laboratory and field evaluations of exotic parasitoids of *Bemisia tabaci* (Gennadius) (Biotype "B") (Homoptera: Aleyrodidae) in the Lower Rio Grande Valley of Texas. Biol. Control. 12: 127-135.

Goolsby, J. A., P. J. DeBarro, A. A. Kirk, R. W. Sutherst, L. Canas, M. A. Ciomperlik, P. C. Ellsworth, J. R. Gould, D. M. Hartley, K. A. Hoelmer, S. E. Naranjo, M. Rose, W. J. Roltsch, R. A. Ruiz, C. H. Pickett, D. C. Vacek. 2005. Post-release evaluation of biological control of *Bemisia tabaci* biotype "B" in the USA and the development of predictive tools to guide introductions for other countries. Biol. Control. 32: 70-77.

Graham, O., and J. Hourrigan. 1977. Eradication programs for arthropod parasites of livestock. J. Med. Entomol. 13: 629-658.

Greenberg, S. M., T-X. Liu, and J. J. Adamczyk. 2009. Thrips (Thysanoptera: Thripidae) on cotton in the Lower Rio Grande Valley of Texas: species composition, seasonal abundance, damage, and control. Southwest. Entomol. 34: 417-430.

Guenther, J., G. Greenway, and J. Goolsby. 2012. Use and costs of insecticides to control Zebra Chip and psyllids. Southwest. Entomol. 37: 263-270.

Guo, J., S. Hatt, K. He, J. Chen, F. Francis, and Z. Wang. 2017. Nine facultative endosymbionts in aphids. A review. J. Asia-Pac. Entomol. 20: 794 –801. doi: 10.1016/j.aspen.2017.03.025

Guzman-Cornejo, C., R.G. Robbins, A.A. Guglielmone, G. Montiel-Parra and T.M. Perez. 2011. The Amblyomma (Acari: Ixodida: Ixodidae) of Mexico: identification keys, distribution and hosts. Zootaxa 2998: 16-38.

Harcourt, D.G., 1963. Major mortality factors in the population dynamics of the diamondback moth, Plutella maculipennis (Curt.) (Lepidoptera: Plutellidae). Mem. Ent. Soc. Can., 95: 55-66. https:// doi.org/10.4039/entm9532055-1

Henneberry T. J., and R. M. Faust. 2008. Introduction. In: Gould J. et al. (eds) Classical biological control of *Bemisia tabaci* in the United States. Springer, Berlin.

Hogenhout S. A., E. D. Ammar, A. E. Whitfield, and M. G. Redinbaugh. 2008. Insect vector interactions with persistently transmitted viruses. Annu. Rev. Phytopathol. 46:327–359.

Horowitz, A. R., M. Ghanim, E. Roditakis, R. Nauen, and I. Ishaaya. 2020. Insecticide resistance and its management in *Bemisia tabaci* species. J. of Pest Science. https://doi-org.srv-proxy2.library.tamu.edu/10.1007/s10340-020-01210-0

Hinomoto, N., M. Osakabe, T. Gotoh, and A. Takafuji. 2001. Phylogenetic analysis of green and red forms of the two-spotted spider mite, *Tetranychus urticae* Koch (Acari: Tetranychidae), in Japan, based on mitochondrial cytochrome oxidase subunit 1 sequences. Appl. Entomol. Zool. 36: 459-464.

Hoberlandt, L. 1955. Results of the Zoological Scientific Expedition of the National Museum in Praha to Turkey: 18. Hemiptera IV, Terrestrial Hemiptera-Heteroptera of Turkey.

Hopkins, B. W., A. E. Knutson, J. S. Bernal, M. F. Treacy, and C. W. Smith. 2009. Species composition, damage potential, and insecticide susceptibility of stink bugs in cotton in the Lower Gulf Coast region of Texas. Southwest. Entomol. 35: 19-32.

Horowitz, A. R., M. Ghanim, E. Roditakis, R. Nauen, and I. Ishaava. 2020. Insecticide resistance and its management in *Bemisia tabaci* species. J. Pest Sci. DOI: 10.1007/s10340-020-01210-0

Hooker, W. A., F. C. Bishop, and H. P. Wood. 1912. The life history and bionomics of some North American ticks. USDA Bull. 106. US Gov. Print Off. Wash. D.C. 239 pp.

Hsiao, T.H. 1988. Host specificity, seasonality and bionomics of Leptinotarsa beetles. In: Jolivet P., E. Petitpierre, and T. H. Hsiao (eds) Biology of Chrysomelidae. Series Entomologica, vol 42. Springer, Dordrecht

Isakeit, T. "Cotton blue disease, a virus disease not in Texas, so far. Texas Row Crops Newsletter, July 2019. 10 October 2019. <https://agrilife.org/texasrowcrops/2019/07/03/cotton-blue-disease-a-virus-disease-not-in-texas-so-far/>

Jasrotia, P., S. Katare, P. L. Kashyap, S. Kumar, A. S. Kharub, and G. P. Singh. 2019. Efficacy of few selected insecticides for the management of foliar aphid complex in barley. Wheat and Barley Research. 10: 228–230. doi: 10.25174/2249-4065/2018/83632

Jones, D. R. 2003. Plant viruses transmitted by whiteflies. Eur. J. Plant Pathol. 109:195–219.

Kariyat, R. R., and J. Chavana. 2018. Field data on plant growth and insect damage on the noxious weed *Solanum eleaegnifolium* in an unexplored native range. Data in brief, 19, 2348–2351. doi:10.1016/j.dib.2018.07.022

Kariyat, R. R., I. Gaffoor, S. Sattar, C. W. Dixon, N. Frock, J. Moen, C. M. De Moraes, M. C. Mescher, G. A. Thompson, and S. Chopra. 2019. Sorghum 3-Deoxyanthocyanidin Flavonoids Confer Resistance against Corn Leaf

Aphid. J. Chem. Ecol. 45: 502-514.

Kaur, J., J. Chavana, P. G. Soti, A. Racelis, and R. R. Kariyat. 2020. Arbuscular Mycorrhizal Fungi (AMF) influences growth and insect community dynamics in Sorghum-sudangrass (*Sorghum x drummondii*). Arthropod-Plant Inte. 14:301-315.https://doi.org/10.1007/s11829-020-09747-8

Kerns, D. L., S. Brown, J. Beuzelin, and K. M. Guidry. 2015. Sugarcane aphid: a new invasive pest of sorghum. Louisiana Agri. 58: 12-14.

Knutson, A., R. D. Bowling, M. Brewer, E. Bynum, and P. Porter. 2016. The sugarcane aphid: management guidelines for grain and forage sorghum in Texas. Texas A&M Agrilife Extension, NTO-035.

Kocan, K. M., J. A. Hair, S. A. Ewing and J. G. Stratton. 1992. Transmission of *Anaplasma marginale* Theiler by *Dermacentor andersoni* Stiles and *Dermacentor variabilis* (Say). American J. Vet. Res. 42: 15-18.

Lada, P., S. Nava, L., Mendoza-Uribe, A. G. Caceres, M. J. Delgado-de al, M. B. Labruna, L. A. Durden, M. E. J. Allerdice, C. D. Paddock, M. P. J. Szabo, J. M. Venzal, A. A. Guglielmone, and L. Beati. 2018. The *Amblyomma maculatum* Koch, 1844 (Acari: Ixodidae) group of ticks: phenotypic plasticity or incipient speciation. Parasite. Vector. 11: 610-632.

Lefoe G, L. Haegi, L. Rumpff, D. Gopurenko, A. T. Slater, K. Butler, and C. E. Hauser. 2020. Assessing the fundamental host-range of *Leptinotarsa texana* Schaeffer as an essential precursor to biological control risk analysis. Biol. Control. 143: 104165. https://doi.org/10.1016/j.biocontrol.2019.104165.

Leigh, T. F. 1961. Insecticidal susceptibility of *Nysius raphanus*, a pest of cotton. J. Econ. Entomol. 54: 120-121.

Louis, J., S. Basu, S. Varsani, L. Castano-Duque, V. Jiang, W. P. Williams, G. Felton, and D. S. Luthe. 2015. Ethylene Contributes to maize insect resistance1-Mediated Maize Defense against the Phloem Sap-Sucking Corn Leaf Aphid. Plant Physiol. 169: 313–324. doi: 10.1104/pp.15.00958

Martin, E., M. K. Borucki, J. Thissen, S. Garcia-Luna, M. Hwang, M. Wise de Valdez, C. J. Jaing, G. L. Hamer, and M. Frank. 2019. Mosquito-borne viruses and insect-specific viruses revealed in field-collected mosquitoes by a monitoring tool adapted from a microbial detection array. Appl. Environ. Microbiol. 85: **DOI:** 10.1128/AEM.01202-19

Metcalf, C. L., W. P. Flint, and R. L. Metcalf. 1962. "Destructive and Useful Insects," 4th ed. pp. 582-587. McGraw-Hill, New York. PATTON, R. L. 1963.

Michaud, J. P., R. J. Whitworth, H. Schwarting, B. McCornack, and S. Zukoff. 2016. Sorghum insect management. Kansas State University Research and Extension. MF742.

Michels, G. J., and J. H. Matis. 2008. Corn leaf aphid, *Rhopalosiphum maidis* (Hemiptera: Aphididae), is a key to greenbug, *Schizaphis graminum* (Hemiptera: Aphididae), biological control in grain sorghum, *Sorghum bicolor*. Eur. J. Entomol. 105: 513-520.

Mitchell, E.A., P.C. Williamson, P.M. Billingsley, E.E. Ferguson, and M.S. Allen. 2016. Frequency and distribution of Rickettsiae, Borreliae, and Ehrlichiae detected in human parasitizing ticks, Texas USA. Emerg. Infect. Dis. 22: 312-315.

Moraes, J. C., M. M. Goussain, G. A. Carvalho, and R. R. Costa. 2005. Feeding non-preference of the corn leaf aphid *Rhopalosiphum maidis* (Fitch, 1856) (Hemiptera: Aphididae) to corn plants *(Zea mays* L.) treated with silicon. Ciênc. Agrotec. 29: 761-766.

Munyaneza, J. E., J. A. Goolsby, J. M. Crosslin, and J. E. Upton. 2007. Further evidence that zebra chip potato disease in the lower Rio Grande Valley of Texas is associated with *Bactericera cockerelli*. Subtrop. Plant Sci. 59:30-37.

National Cotton Council of America. 2018. Eradication and Economic and Environmental Benefits. 12 Oct. 2019. <https://www.cotton.org/tech/pest/bollweevil/eradication4.cfm>

Nava, S., L. Beati, M. B. Labruna, A. G. Cáceres, A. J. Mangold, A. A. Guglielmone. 2014. Reassessment of the taxonomic status of *Amblyomma cajennense* (Fabricius 1787) with the description of three new species, *Amblyomma tonelliae* n. sp., *Amblyomma interandinum* n. sp., and *Amblyomma patinoi* n. sp. and reinstatement of *Amblyomma mixtum* Koch 1844, and *Amblyomma sculptum* Berlese, 1888 (Ixodida: Ixodidae). Ticks Tick-Borne Dis. 5: 252-276.

Olckers, T., H. G. Zimmermann, and J. H. Hoffmann (n.d.). 1995. Interpreting ambiguous results of host-specificity tests in biological control of weeds: assessment of two Leptinotarsa species (Chrysomelidae) for the control of *Solanum elaeagnifolium* (Solanaceae) in South Africa. Biol. Control. *5*: 336–344.

Olckers T., J. H. Hoffmann, V. C. Morán, F. A. C. Impson, and M. P.Hill. 1999. The initiation of biological control programmes against *Solanum elaeagnifolium* Cavanilles and *S. sisymbriifolium* Lamarck (Solanaceae) in South Africa. Afr. Entomol. 55–63. January 8 2020. <https://pdfs.semanticscholar.org/7164/946f7fa351776560170d0df12c2daeb2f6df.pdf>

Pallister, J. C. 1953. The leaf beetles of North Central Mexico collected on the David Rockefeller Mexican Expedition (Coleoptera, Chrysomelidae). America Museum Novitates 1623.

Parker, R. R., G. M. Kohls, G. W. Cox, and G. E. Davis. 1939. Observation on an infectious agent from *Amblyomma maculatum*. Public Health Rept. 54: 1482-1484.

Pepin, K.M., R. J. Eisen, P.S. Mead, J. Piesman, D. Fish, A.G. Hoen, A. G. Barbour, S. Hamer, and M. A. Diuk-Wasser. 2012. Geographic variation in the relationship between human Lyme disease incidence and density of infected host-seeking *Ixodes scapularis* nymphs in the Eastern United States. Am. J. Trop. Med. Hyg., 86: 1062–1071.

Perez de Leon, A.A., D. Teel, A. N. Auclair, M. T. Messenger, F. Guerrero, G. Schuster, and R. J. Miller. 2012. Integrated strategy for sustainable cattle fever tick eradication in U.S.A. is required to mitigate the impact of global change. Front. Physiol. 3: 1-17.

Petanidou T., M. V. Price, N. M. Waser, A. Kantsa, T. Tscheulin, R. R. Kariyat, N. Krigas, M. C. Mescher, C. M. De Moraes and J. L. Bronstein. 2018. Pollination and reproduction of an invasive plant inside and outside its ancestral range. *Acta Oecologica* 89, 11-20.

Philips C. R., Z. Fu, T. P. Kuhar, A. M. Shelton, and R. J. Cordero. 2014. Natural history, ecology, and management of diamondback moth (Lepidoptera: Plutellidae), with emphasis on the United States. J. Integr. Pest Manag. 5: D1-D11.

Pitre, H. N. 1967. Greenhouse studies of the host range of Dalbulus maidis, a vector of the corn stunt virus. J. Econ. Entomol. 60: 417-421.

Nault, L. R., and O. E. Bradfute. 1979. Corn stunt: involvement of a complex of leafhopper-borne pathogens. Pp. 561-586. In: Leafhopper vectors and plant disease agents. K. Maramorosch and K. F. Harris (eds.). Academic Press, New York, NY. 654 pp.

Nault, L. R., and L. V. Madden. 1985. Ecological strategies of Dalbulus leafhoppers. Ecolog. Entomol. 10: 57-63.

Pallister, J. C. 1953. The leaf beetles of North Central Mexico collected on the David Rockefeller Mexican Expedition (Coleoptera, Chrysomelidae). America Museum Novitates. 1623.

Riley, D. G., V. J. Shimat, S. Rajagopalbabu, and D. Stanley. 2011. Thrips vectors of tospoviruses. J. Integr. Pest Manag. 2: 1-10.

Saksena, K. N., S. R. Singh, and W. H. Sill. 1964. Transmission of Barley Yellow-Dwarf Virus by four biotypes of the corn leaf aphid, *Rhopalosiphum maidis*. J. Econ. Entomol. 57: 569-571.

Sames, W. J. 4th, R. Bueno Jr., J. Hayes, J. K. Olson. 1996. Insecticide susceptibility of Aedes aegypti and Aedes albopictus in the Lower Rio Grande Valley of Texas and Mexico. J.Am. Mosquito Contr. 12:487-490.

Sanchez, J. A., M. La-Spina, and A. Lacasa. 2014. Numerical response of *Nesidiocoris tenuis* (Hemiptera: Miridae) preying on *Tuta absoluta* (Lepidoptera: Gelechiidae) in tomato crops. Eur. J. Entomol. 111: 387-395.

Sappington, T. W., L. S. Hesler, K. C. Allen, R. G. Luttrell, and S. K. Papiernik. 2018. Prevalence of sporadic insect pests of seedling corn and factors affecting risk of infestation. J. Integr. Pest Manag. 9: 1-27.

Scoles, G.A., H.J. Hutcheson, J.L. Schlater, S.G. Hennaager, A.M. Pelzel and D.P. Knowles. 2011. Equine piroplasmosis associated with *Am blyomma cajennense* ticks, Texas, USA. Emerg. Infect. Dis. 17: 1903-1904.

Schwint, O.N., D.P. Knowles, M.W. Ueti, L.S. Kappmeyer, & G.A. Scoles. 2008. Transmission of *Babesia caballi* by *Dermacentor nitens* (Acari: Ixodidae)is restricted to one generation in the absence of alimentary reinfection on a susceptible equine host. J. Med. Entomol. 45: 1152-1155.

Shelton, A. M., F. V. Sances, J. Hawley, J. D. Tang, M. Boune, D. Jungers, H. L. Collins, and J. Farias. 2000. Assessment of insecticide resistance after the outbreak of diamondback moth (Lepidoptera: Plutellidae) in California in 1997. J. Econ. Entomol. 93: 931–936.

Silva, T. F., R. L. Corrêa, Y. Castilho, P. Silvie, J-L Bélot, and M.F.A. Vaslin. 2008. Widespread distribution and a new recombinant species of Brazilian virus associated with cotton blue disease. Virol. J. 123: https://doi.org/10.1186/1743-422X-5-123

Singh, N.K., J. A. Goolsby, D. I. Shapiro-Ilan, R. J. Miller, M. Setamou, and A. A. Perez de Leon. 2018. Effect of immersion time on efficacy of entomopathogenic nematodes against engorged females of cattle fever tick, *Rhipicephalus* (*Boophilus*) *microplus.* Southwest. Entomol. 43: 19-28.

Smith, J.W. 1998. Boll weevil eradication: area-wide pest management. Annu. Entomol. Soc. of Am. 91: 239-247.

Sonenshine, D. 2018. Range expansion of tick disease vectors in North America: implications for spread of tick borne diseases. Int. J. Environ. Res. and Public Health 15: 478-486.

Sosa-Gómez, D. R., B. S. Corrêa-Ferreira, B. Kraemer, A. Pasini, P. E. Husch, C. E. Delfino Vieira, C. B. Reis Martinez, and I. O. Negrão Lopes. 2019. Prevalence, damage, management and insecticide resistance of stink bug populations (Hemiptera: Pentatomidae) in commodity crops. Agric. For. Entomol. DOI: 10.1111/afe.12366

Sperling, J. L. Haley, and B. K. Mitchell. 1991. A comparative study of host recognition and the sense of taste in Leptinotarsa. J. Exp. Biol. 157: 439-459.

Stiller, D., and M. E. Coan. 1995. Recent developments in elucidating tick vector relationships for anaplasmosis and equine piroplasmosis Vet. Parasitol. 57: 97-108.

Strickland, R.K., R.R. Gerrish, J.l. Hourrigan and G.O. Schubert. 1976. Ticks of Veterinary Importance. USDA Agricultural Handbook No. 485. U. S. Gov. Print. Off. Wash. D.C. 122 pp.

Syzoni, B., I. Srinath, M. Esteve-Gassent, B. Lupiani. and R. Ivanek. 2015. Exploratory spatial analysis of Lyme disease in Texas – what can we learn from the reported cases? Biomed Central Public Health 15: 924.

Teel, P.D., H.R. Ketchum, D.E. Mock, R.E. Wright and O.F. Strey. 2010. The gulf coast tick: a review of the life history, ecology, distribution, and emergence as an arthropod of medical and veterinary importance. J. Med. Entomol. 47: 707-722

Texas Boll Weevil Eradication Foundation, Inc. 12 Oct. 2019. <https://www.txbollweevil.org/index.php>

Tidwell, J., C.J. Vitek, D.B. Thomas and J.M. Thomas. 2019. Status update on the threat of Babesiosis returning to the United States. J. Dairy Vet. Sci. 7: 1-4.

Torres, J. B., and A. F. Bueno. 2018. Conservation biological control using selective insecticides a valuable tool for IPM. Biol. Contol. 126: 53–64.U.S. Department of Health of Agriculture. 1960. National healthcare quality report 2012 (Report No. 1). Plant Pest Control Division Agricultural Research Service. Agency for Healthcare Research and Quality.

United States Department of Agriculture, Texas Cattle Fever. Special Collections. 30 January 2020. <www.nal.usda.gov/exhibits/speccoll/exhibits/show/parasitic-diseases-with-econom/parasitic-diseases-with-econom/texas-cattle-fever.>

United States Department of Agriculture, Texas Field Office. 2019. National agricultural statistics service, Texas county estimates. 15 January 2020. <https://www.nass.usda.gov/Statistics_by_State/Texas/Publications/County_Estimates/ce_pdf/index.php>

U.S. Department of Health of Agriculture. 1960. National healthcare quality report (Report No. 1). Plant Pest Control Division Agricultural Research Service. Agency for Healthcare Research and Quality. Cooperative Economic Insect Report, Volume 10.

Urbaneja, A., H. Montón, and O. Molla. 2009. Suitability of the tomato borer *Tuta absoluta* as prey for *Macrolophus pygmaeus* and *Nesidiocoris tenuis*. J. Appl. Entomol. 133: 292-296.

Vinson, S. B., and T. A. Scarborough. 1991. Interactions between *Solenopsis invicta* (Hymenoptera: Formicidae), *Rhopalosiphum maidis* (Homoptera: Aphididae), and the Parasitoid *Lysiphlebus testaceipes* Cresson (Hymenoptera: Aphidiidae). Ann. Entomol.Soc. Amer.. 84: 158–164. doi: 10.1093/aesa/84.2.158

Vyavhare, S. and D. Kerns. 2017a. Cotton Aphids. Texas A&M Agrilife Extension, ENTO-074.

Vyavhare S. and D. Kerns. 2017b. Thrips. Texas A&M University Ento-069.

Vyavhare, S. S., M. O. Way, and R. F. Medina. 2014. Stink bug species composition and relative abundance of the redbanded stink bug (Hemiptera: Pentatomidae) in soybean in the upper gulf coast Texas. Environ. Entomol. 43: 1621-1627.

Vyavhare, S., D. Kerns, C. Allen, R. Bowling, M. Brewer, and M. Parajulee. 2018. Managing Cotton Insects in Texas. Texas A&M Agrilife Extension, ENTO-075.

Waldrup, K. A., J. Moritz, D. Baggett, S. Magyar, and G. G. Wagner. 1992. Monthly incidence of *Theileria cervi* and seroconversion to *Babesia odocoilei* in white-tailed deer (*Odocoileus virginianus*) in Texas. J. Wildl. Dis. 28:457-459.

Weatherspark. 2019 Report - Average weather at Rio Grande Valley International Airport. 15 January 2020. <https://weatherspark.com/y/145897/Average-Weather-at-Rio-Grande-Valley-International-Airport-Texas-United-States-Year-Round>

Wene, G. P., and L. W. Sheets. 1964. Notes on control of stink bugs affecting cotton in Arizona. J. Econ. Entomol. 57: 60-62.

Whitworth, R. J., and H. Davis. 2009. Crop Pests – Stink Bugs. Kansas State University. MF-2891.

Williamson, P. C., P. M. Billingsley, G. J. Teltow, J. P. Seals, M. A. Turnbough, and S. F. Atkinson. 2010. *Borrelia, Ehrlichia* and *Rickettsia* spp. in ticks removed from persons, Texas, USA. Emerg. Infect. Dis.16: 441-446.

Wilson, B. E. 2019. Hemipteran Pests of Sugarcane in North America. Insects. 10: 107.

Wood, E. A., Jr., and K. J. Starks. 1972. Damage to sorghum by a lygaeid bug, *Nysius raphanus*. J. Econ. Entomol. 65: 1507-1508.

Workneh, F., L. Paetzold, A. Silva, C. Johnson, A. Rashed, I. Badillo-Vargas, N. C. Gudmestad, and C. M. Rush. 2018. Assessments of temporal variations in haplotypes of 'Candidatus Liberibacter solanacearum' and its vector, the potato psyllid, in potato fields and native vegetation. Environ. Entomol. 47: 1184-1193.

Zhang, L., S. M. Greenberg, Y. Zhang, and T-X Liu. 2011. Effectiveness of thiamethoxam and imidacloprid seed treatments against *Bemisia tabaci* (Hemiptera: Aleyrodidae) on cotton. Pest Manag. Sci. 67: 226-232.

Zhang, X., M.I. Meltzer, C.A. Peña, A.B. Hopkins, L. Wroth and A.D. Fix. 2006. Economic impact of Lyme Disease. Emerging Infect. Dis. 12: 653-660.