
An Overview of Chilli Thrips, *Scirtothrips dorsalis* (Thysanoptera: Thripidae) Biology, Distribution and Management

Vivek Kumar, Garima Kakkar, Cindy L. McKenzie,
Dakshina R. Seal and Lance S. Osborne

Additional information is available at the end of the chapter

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1. Introduction

The industrial revolution, globalization and international trade liberalization are some of the important events that have afforded vast opportunities for invasive insect species to establish in new territories [1]. These invasive species, facing no challenge by their natural enemies, thrive well in the new environment [2]. In addition to the disturbance they cause to the biodiversity, pest invasion in any country results in increased pressure on biosecurity, national economy, and human health management systems [1, 3, 4]. Apart from economic loss in managing them, these pests pose a significant detrimental impact on tourism and recreational value of the region, which further adds in indirect economic damage to the nation [5]. Of this large group of invasive pests, thrips are one of the most important members. The invasive status gained by thrips across the globe is due to their high degree of polyphagy, wide host range and easy dispersal that can be anthropogenic or natural (wind-mediated).

The earliest fossil record of order Thysanoptera dates back to the Late Triassic period, from the state of Virginia in the United States and the country Kazakhstan in Central Asia, but their abundance was rare until the Cretaceous period from which many specimens of Thysanoptera have been recorded [6]. The order Thysanoptera was given its current taxonomic rank by an Irish entomologist, A. H. Haliday in 1836, and since then more than 8,000 species of thrips have been reported. In this insect order, the genus *Scirtothrips* Shull contains more than 100 thrips species, among which 10 species have been reported as serious pests of agricultural crops [7]. Within this genus, *Scirtothrips dorsalis* Hood is a significant pest of various economically important vegetable, ornamental and fruit crops in southern and eastern Asia, Oceania and

parts of Africa [8, 9]. *S. dorsalis* is native to the Indian subcontinent and is a polyphagous pest with more than 100 reported hosts among 40 different families of plants [10]. However, in the past two decades, increased globalization and open agricultural trade has resulted in the vast expansion of the geographical distribution and host range of the pest. In the United States, it is a new invasive pest where the first established population of *S. dorsalis* was reported in 2005 from Florida. Since then it has emerged as a serious pest of various economically important host crops in the southeastern regions of the United States. It has been reported from 30 counties in Florida, 8 counties in Texas with several positive reports of its invasion from Alabama, Louisiana, Georgia, and New York. In a recent study [11], this pest was found attacking 11 different hosts at a fruit nursery in Homestead, Florida. Interestingly, they were found to reproduce on nine plant taxa that had not previously been reported as host plants in the literature suggesting that the host range of this insidious pest is continuing to expand as it invades new regions. The small size and cryptic nature of adults and larvae enables *S. dorsalis* to inhabit microhabitats of a plant often making monitoring and the identification difficult. *S. dorsalis*' life stages may occur on meristems and other tender tissues of all above ground parts of host plants [12]. Consequently, the opportunity of trans-boundary transportation of *S. dorsalis* through the trade of plant materials is high [13]. Existence of any variation in phenotypic and genetic makeup of such a pest makes identification much more difficult [14].

This chapter is intended to summarize the parameters facilitating worldwide distribution of this pest, damage potential and the advancement in the post-invasion management strategies being practiced in the United States and other parts of the world. The focus will be on the latest development in the integrated pest management of *S. dorsalis* including identification techniques and biological, chemical and cultural control strategies.

2. Background information

The great reproductive potential and keen ability for invasion combined with easy adaptation to newly invaded areas are a few of the qualities which make *Scirtothrips* species major concerns for agriculture in many countries [15]. From the beginning, *S. dorsalis* has been reported as an opportunistic generalist species that is able to feed on a variety of host plants, depending upon availability in the region of incidence. The first reference to *S. dorsalis* was in early 1900's when it was reported damaging the tea crop in the Tocklai area of Assam state in India. In later years *S. dorsalis* was responsible for damaging the tea crops in all of the major tea growing regions of eastern India including Cachar, the Assam Valley, Terai and the Dooars [16]. In 1916, this pest was reported infesting castor in the Coimbatore district of the southern part of India and later was found infesting other hosts in the region including chilli, groundnuts, mango, beans, cotton, brinjal (eggplant) and *Casia fistula* [17, 18]. Young leaves, buds, and tender stems of the host plants were severely damaged. Thrips repeated puncturing of tender leaf tissues with their stylet produces 'sandy paper lines' on the epidermis of the leaves and eventual crinkling of leaves. In India, the characteristic leaf curl damage caused by this pest is known as "Murda" (Hindi meaning- dead body) disease, because infestations resulted in the death of plants [19]. Many different scientific names have been assigned to *S. dorsalis* since it was first described in

1919, mainly because of the lack of sufficient scientific literature regarding morphological differences and variations in host range from the different geographical regions. During the last 100 years, the host range and the bio-geographical range of *S. dorsalis* have broadened. The thrips is established in all of the habitable continents except Europe, where repeated introductions have been intercepted and eliminated [13]. Studying the history of *S. dorsalis* aids in the understanding of behavioral and morphological diversity exhibited by this species as a result of biological and ecological variations that have occurred during its long migration to different parts of the world.

3. Geographical distribution

3.1. Worldwide distribution

S. dorsalis is widely distributed along its native range in Asia including Bangladesh, Brunei Darussalam, China, Hong Kong, India, Indonesia, Japan, Republic of Korea, Malaysia, Myanmar, Pakistan, Philippines, Sri Lanka, Taiwan, and Thailand. Further south *S. dorsalis* occurs in northern Australia and the Solomon Islands. On the African continent, the pest is reported from South Africa and the Ivory Coast, with plant health quarantine interceptions suggesting a wider distribution across West Africa and East Africa (Kenya) [20]. *S. dorsalis* is in Israel as well as in the Caribbean including Jamaica, St. Vincent, St. Lucia, Barbados and Trinidad [12]. In South America, *S. dorsalis* has been found causing serious damage to grapevine in western Venezuela [20].

3.2. U.S. invasion

Changing climatic conditions and globalization have resulted in the increasing importance of invasive species as recurrent problems around the globe. More than 50,000 non-indigenous species have already been introduced into the United States, causing an estimated annual damage of more than \$120 billion in forestry, agriculture and other sectors of society [3, 21]. The rich vegetation and neotropical climate of Florida make the state suitable for the invasion and establishment of exotic flora and fauna [22]. *S. dorsalis* is a newly introduced insect pest in Florida believed to have originated from Southeast Asia. In between 1984-2002, it was intercepted about 89 times by USDA-APHIS inspectors at various US ports-of-entry [23]. Most of the records of interception were from imported plant materials including cut flowers, fruits and vegetables. With the exception of Hawaii, the presence of this tropical south Asian pest was not confirmed in the Western Hemisphere until 2003. In Florida, *S. dorsalis* was reported from Okeechobee County in 1991 and from Highland County in 1994 but failed to establish a durable population [24]. In 2003, Tom Skarlinsky (USDA-APHIS-PPQ) reported live larvae and pupae under the calyx of treated peppers in a shipment of *Capsicum spp.* traced back to hot pepper production areas in St. Vincent and the Grenadines, West Indies [25]. Later, with the collaborative efforts of the USDA (APHIS) and the Institute of Food and Agricultural Sciences (University of Florida), *S. dorsalis* was found established in different agricultural districts of St. Lucia and St. Vincent [26], Barbados, Suriname, Trinidad and Tobago, and

Venezuela [25]. In 2005, *S. dorsalis* was found on pepper and 'Knockout' Rose plants in retail garden centers in Florida and Texas. Subsequently, *S. dorsalis* has been reported many times on different ornamental plants in commercial nurseries throughout Florida [27]. In a collaborative survey over a two-month period (Oct-Nov 2005), the Florida Department of Agricultural and Consumer Services (FDACS) and the University of Florida found infestations 77 times in 16 counties [25]. Of the 77 positive observations, 66 were found on roses, 10 on *Capsicum* and one on *Illicium*.

Venette and Davis [28] projected the potential geographic distribution of *S. dorsalis* in North America. Based on this *S. dorsalis* could extend from southern Florida to the Canadian border, as well as to Puerto Rico and the entire Caribbean region which suggests that this pest could also become widely established in South America and Central America. The small size (< 2 mm in length) and thigmotactic behavior of *S. dorsalis* make it difficult to detect the pest in fresh vegetation, thus, increasing the likelihood of the transportation of the pest through international trade of botanicals. The major pathways of trans-boundary movement of *S. dorsalis* includes (i) air passengers and crew, their baggage, and air cargo of plant propagative materials and fresh ornamentals, fruits, and vegetables, (ii) mail, including mail from express mail carriers, (iii) infested smuggled fresh plant materials, and (4) windborne dispersal [29].

4. Economic impact

Among 8,800 species of thrips, around 5,000 species has been well described with their diverse life history and habitats [6]. Approximately 1% of the members of this order have been reported as serious pests by humans owing to various damages which disrupt their life styles [30]. Thrips can reduce yield or value of the crop directly by using them as food and oviposition site and indirectly by transmitting plant diseases. Their infestation can negatively impact global trade due to the quarantine risks associated with several species in the order. The majority of scientific literature related to economics of thrips deals with four important thrips species: *Thrips tabaci* Lindeman, *T. palmi* Karny, *Frankliniella occidentalis* (Pergande) and *S. dorsalis* [30].

India is one of the world's largest chilli (*Capsicum annum* L.) producers which contributes about 36% (0.45 million tons) of global production [31]. According to a survey by the Asian Vegetable Research and Development Committee, *S. dorsalis* is one of the most important limiting factors for the chilli production in the country along with aphid species *Myzus persicae* Sulzer, *Aphis gossypii* Glover and mite *Polyphagotarsonemus latus* Banks [32]. Yield loss solely dedicated to *S. dorsalis* damage can range between 61 to 74% [33]. Because of its damage potential to chilli pepper, this dreadful pest is commonly referred as chilli thrips.

Globally, the popularity and demand for mango (*Mangifera indica* L.) and its processed product is rising which has resulted in the expansion of area under mango cultivation. Asian countries contribute around 77% of the global mango production followed by Americas (13%) and Africa (9%). In 2005, world mango production was reported as 28.5 million metric tons [34]. Malaysia, which is a major mango consumer (10th largest mango importer in the world), produces mango

on over 4,565 ha [35]. However, their domestic mango production has been reported to suffer considerable economic losses due to thrips infestation with the major thrips species responsible for damage of mango panicle reported as *Thrips hawaensis* Morgan and *S. dorsalis* [36]. Along the same international theme, *S. dorsalis* is considered as a major economic threat to grape and citrus production in Japan [37, 38] and vegetable production in China and US [39].

The invasion of *S. dorsalis* into the Caribbean region prompted an economic analysis to be conducted in 2003 by the United States Department of Agriculture on 28 potential hosts of the pest which suggested that a 5% loss of these crops may lead to a loss value of \$3 billion to the US economy [40]. Assessment of the damage potential of *S. dorsalis* from Florida's perspective as the port of entry showed that there is an immediate need for development of effective management practices against this pest. In 2010, the US horticulture (greenhouse/nursery) industry contributed approximately \$15.5 billion to the US economy, among which Florida was the second largest contributor after California by adding 11.2% to the economy [41]. Florida received cash receipts of approximately \$7.80 billion in 2010 from all agricultural commodities among which top three contributors were greenhouse/nursery, orange and tomato which added about 53% of all cash receipts. Strawberries (\$362 million), peppers (\$295 million), peanuts (\$89 million), cucumbers (\$88 million), cotton (\$49 million), and blueberries (\$47 million) contributed an additional 1 billion (approx.) to Florida's economy. Since these crops have potential to serve as host plants of *S. dorsalis*, even 10% loss of these commodities can cause significant impact on Florida's economy and may open the market for foreign trade [42]. Florida Nurserymen and Growers Association consider *S. dorsalis* as one of the thirteen most dangerous, exotic pests threatening the industry [43].

5. Host plants

Prior to the introduction of *S. dorsalis* into the New World, the host range of this pest included more than 100 plant taxa among 40 families [10]. Subsequent to the introduction of *S. dorsalis* into the New World, the pest was found to attack additional taxa of plants [28]. The main wild host plants belong to the family Fabaceae, which includes Acacia, Brownea, Mimosa and Saraca. In its native range of the Indian subcontinent, chilli crops are reported to be attacked by 25 different pests, among which *S. dorsalis* is considered as one of the most serious threats [44]. *S. dorsalis* is also abundant on *Arachis* in India [45], sacred lotus in Thailand [10], and tea and citrus in Japan [46]. Among the potential economic hosts of this pest listed by Venette and Davis [28] are banana, bean, cashew, castor, citrus, cocoa, corn, cotton, eggplant, grapes, kiwi, litchi, longan, mango, melon, peanut, pepper, poplar, rose, strawberry, sweet potato, tea, tobacco, tomato, and wild yams (*Dioscorea spp.*). Interestingly, *S. dorsalis* is not reported reproducing on all of the hosts mentioned in the literature and plant species has been designated as a host plant based on the presence of adult thrips. While *S. dorsalis* is known to forage on wide range of plant species, a true host must be identified by its ability to support thrips reproduction in addition to provisioning food and shelter. Based on information obtained from the Global Pest and Disease Database [47], *S. dorsalis* was reported to feed on (not necessarily

reproduce on) more than 225 plant taxa worldwide in 72 different families and 32 orders of plants. In Florida, *S. dorsalis* has been reported from 61 different plants till 2011 (Table 1). Disparities in host selection in different geographical regions are documented in the literature. For example, *S. dorsalis* is reported on mango in Puerto Rico but not in adjacent Caribbean islands where it was reported earlier on other host plants. *S. dorsalis* is a significant pest of citrus in Japan [48] and Taiwan [49], but not in India or the United States. Many factors could be attributed to the differences in host plants of *S. dorsalis* reported from different geographical regions. These various factors could include variation in competition with other pests, availability of predators in the region of invasion, availability of hosts, environmental conditions, etc. [42], but could also be the result of differential biological activity of different *S. dorsalis* biotypes/cryptic species, none of which have yet been reported.

Scientific name	Common or trade name
<i>Antirrhinum majus</i> L.	Liberty Classic White Snapdragon
<i>Arachis hypogaea</i> L.	Peanut or groundnut
<i>Begonia</i> sp.	Begonia
<i>Breynia nivosa</i> (W. Bull) Small	Snow bush, snow-on-the-mountain
<i>Camellia sinensis</i> (L.) Kuntze	Tea
<i>Capsicum annuum</i> L.	Jalapeno pepper, Bonnet pepper
<i>Capsicum frutescens</i> L.	Chilli pepper
<i>Capsicum</i> spp.	
<i>Celosia argentea</i> L.	Celosia – red fox
<i>Citrus</i> spp.	
<i>Conocarpus erectus</i>	
<i>Coreopsis</i> sp.	Tickseed
<i>Cuphea</i> sp.	Waxweed, tarweed
<i>Duranta erecta</i> L.	golden dewdrop, pigeonberry, skyflower
<i>Euphorbia pulcherrima</i> Willd.	Poinsettia
<i>Eustoma grandiflorum</i> (Raf.) Shinn.	Florida Blue Lisianthus
<i>Ficus elástica</i> 'Burgundy' Roxb. Ex Hornem.	Burgundy Rubber Tree
<i>Gardenia jasminoides</i> J. Ellis	Jasmine
<i>Gaura lindheimeri</i> Engelm. & Gray	Lindheimer's beeblossom
<i>Gerbera jamesonii</i> H. Bolus ex Hook. F.	Gerber daisy
<i>Glandularia x hybrida</i> (Grönland & Rümpler) Neson & Pruski	Verbena
<i>Gossypium hirsutum</i> L.	Cotton
<i>Hedera helix</i> L.	English Ivy
<i>Illicium floridanum</i> Ellis	Florida anisetree

Scientific name	Common or trade name
<i>Impatiens walleriana</i> Hook. F.	Super Elfin White
<i>Jasminum sambac</i> (L.) Ait.	Pikake
<i>Lagerstroemia indica</i> L.	Crape myrtle
<i>Laguncularia recemosa</i> (L.) Gaertn. f.	White buttonwood
<i>Ligustrum japonicum</i> Thunb.	Japanese privet
<i>Litchi chinensis</i> Sonn.	Litchi
<i>Mahonia bealei</i> (Fortune) Carrière	Leatherleaf mahonia
<i>Manilkara zapota</i> (L.) D. Royen	Sapodilla
<i>Mangifera indica</i> L.	Mango
<i>Murraya paniculata</i> (L.) Jack	Orange-jasmine
<i>Ocimum basilicum</i> L.	Sweet Basil
<i>Pelargonium x hortorum</i> Bailey	Geranium
<i>Pentas lanceolata</i> (Forssk.) Deflers	Graffiti White
<i>Persea americana</i> Mill.	Avocado
<i>Petunia x hybrida</i>	Petunia Easy Wave Red
<i>Pittosporum tobira</i> (Thunb.) Ait. f.	Variegated Pittosporum
<i>Plectranthus scutellarioides</i> (L.) R. Br.	Coleus
<i>Pouteria campechiana</i> (Kunth) Baehni	Canistel
<i>Rhaphiolepis indica</i> (L.) Lindl. ex Ker Gawl.	Shi Ban Mu
<i>Ricinus communis</i> L.	Castor Bean
<i>Rhaphiolepis umbellate</i> (Thunb.)	Yeddo Hawthorn
<i>Richardia brasiliensis</i> Gomes	Brazil Pusley
<i>Rhododendron spp.</i>	Azalea
<i>Rosa X 'Radrazz'</i>	'Knockout®' rose
<i>Salvia farinacea</i> Benth.	Victoria blue
<i>Schefflera arbicola</i> (Hayata) Merr.	Dwarf umbrella tree
<i>Strobilanthes dyerianus</i> Mast.	Persian shield
<i>Synsepalum dulcificum</i> (Schumach. & Thonn.) Daniell	Miracle fruit
<i>Tagetes patula</i> L.	Marigold
<i>Tradescatia zebrina</i> hort. ex Bosse	Wandering jew
<i>Vaccinium corymbosum</i> L.	Highbush blueberry
<i>Viburnum odoratissimum</i> var. <i>awabuki</i> (K. Koch) Zabel	Sweet viburnum
<i>Viburnum suspensum</i> Lindl.	Viburnum
<i>Viola x wittrockiana</i> Gams	Wittrock's violet
<i>Vitis vinifera</i> L.	Grapevine
<i>Zinnia elegans</i> Jacq.	Zinnia Profusion White

Table 1. Confirmed plant hosts of *Scirtothrips dorsalis* in Florida. Source: [80].

6. Host damage

S. dorsalis feeding on the meristems, terminals and other tender plant parts of the host plant above the soil surface results in undesirable feeding scars, distortion of leaves, and discoloration of buds, flowers and young fruits. The pest prefers young plant tissue and is not reported to feed on mature host tissues. The piercing and sucking mouthparts of *S. dorsalis* can damage the host plant by extracting the contents of individual epidermal cells, leading to the necrosis of tissue. The color of damaged tissue changes from silvery to brown or black. The appearance of discolored or disfigured plant parts suggests the presence of *S. dorsalis*. Adults and larvae of *S. dorsalis* suck the cell sap of the leaves, causing the leaves to curl upward [50]. Severe infestations of *S. dorsalis* cause the tender leaves and buds to become brittle, resulting in complete defoliation and yield loss. For example, heavy infestations of pepper plants by *S. dorsalis* cause changes in the appearance of plants termed “chilli leaf curl” [51]. On many hosts, the thrips may feed on the upper surfaces of leaves when infestations are high. Infested fruits develop corky tissues [52]. Sometimes, plants infested by *S. dorsalis* appear similar to plants damaged by the feeding of broad mites. Plants infested with *S. dorsalis* may show the following damage symptoms: (i) silvering of the leaf surface, (ii) linear thickening of the leaf lamina, (iii) brown frass markings on the leaves and fruits, (iv) grey to black markings on fruits, often forming a distinct ring of scarred tissue around the apex and (v) fruit distortion and premature senescence and abscission of leaves [53]. Apart from causing direct damage to its host *S. dorsalis* also vectors seven plant viruses including chilli leaf curl virus (CLC), peanut necrosis virus (PBNV), peanut yellow spot virus (PYSV), tobacco streak virus (TSV), watermelon silver mottle virus (WsMoV), capsicum chlorosis virus (CaCV) and melon yellow spot virus (MYSV) [8, 10, 54, 55, 56, 57].

7. Identification

Correct identification is a primary step in the development of sound management practices against a pest. Identification helps in attaining previously reported information against the subject species [58] crucial in planning and implementation of an appropriate biological research strategy. Morphological identification characters of *S. dorsalis* are well studied in the literature due to its economic importance and global distribution. However, due to the small size and morphological similarities within the genus, the identification of unknown specimen to species level is a challenge to non-experts.

Larvae of *S. dorsalis* are creamish white to pale in color. Sizes of the first instars, second instars, and pupae range between 0.37-0.39, 0.68-0.71 and 0.78-0.80 mm, respectively [12]. Morphological identification of *S. dorsalis* larva can be made using the following features [59]): D1 and D2 setae present on the head and abdominal terga IX of larvae are simple and funnel-shaped, respectively. The D1 setae on terga X are funnel shaped. The larval pronotum is reticulated and has 6-7 pairs of pronotal setae. Abdominal segments IV-VII of larvae have a total of 8-12 setae each. The distal two thirds of the fore-femora of larvae possess four funnel shaped setae and the body of larvae possesses granular plaques.

The body of adult *S. dorsalis* is pale yellow in color and bear dark brown antecostal ridges on tergites and sternites. Adults are less than 1.5 mm in length with dark wings. The head is wider than long, bearing closely spaced lineations and a pair of eight segmented antennae with a forked sensorium on each of the third and fourth segments. Dark spots that form incomplete stripes are seen dorsally on the abdomen [12]. Three pairs of ocellar setae are present, the third pair, also known as the interocellar setae (IOS), arises between the two hind ocelli (HO) and is nearly the same size as the two pairs of post ocellar setae (POS) on the head. The pronotum consists of closely spaced horizontal lineation. The pronotal setae (anteroangular, anteromarginal and discal setae) are short and approximately equal in length. The posteromarginal setae-II is broader and 1.5 times longer than the posteromarginal setae-I and III. The posterior half of the metanotum presents longitudinal striations; medially located metanotal setae arise behind the anterior margin and campaniform sensilla are absent. Three discal setae are located on the lateral microtrichial fields of the abdominal tergites and the posteromarginal comb on VIII segment is complete. The shaded forewings are distally lighter in color with posteromarginal straight cilia on the distal half and the first and second veins bear three and two widely spaced setae, respectively. Discal setae are absent on sternites and sternites are covered with rows of microtrichia, excluding the antero-medial region [60, 61].

Using traditional taxonomic keys, adult thrips can be identified to genus, but due to the intraspecific morphological variations in many species, identifying them to species requires substantial expertise [7]. For many taxa of thrips it is impossible to assign an immature to a particular species in the absence of adults [62]). In addition, high levels of variation in the basic biology, life history, host selection, pest status, vector efficiency and resistance to insecticides exist in different thrips species. Misidentification of thrips species can lead to the misapplication of management practices, resulting in wasted money, resources and time [63]. Selection of the wrong biological control agents due to the ambiguous identification of the target pest discourages growers to adopt chemical free pest management strategies. Thus, a rapid, species-specific, developmental-stage non-limiting method for identification of thrips species is of paramount importance to implement appropriate IPM strategies.

Taxonomic characterization of thrips, including *S. dorsalis*, has always been difficult due to their small size and cryptic nature. Thus, it is important to utilize the advantage of other methods of identification including molecular techniques which is not limited by the factors associated with morphological identification [64, 65]. Molecular techniques can be cost effective, rapid, and performed by non-taxonomic experts. Recently a molecular marker (rDNA ITS2) has been developed for species specific identification of *S. dorsalis* specimens [65]. However, misidentification of specimens using solely molecular identification based on genetic information available in databases such as Genbank and EMBL is very common [66] until a voucher specimen or photo-documentation is available to confirm the identity. Thus, it is important to integrate both identification methods (morphological + molecular) to achieve a double confirmation system for validating identification of various thrips species using a single specimen. There are a few such techniques available such as sonication of specimens for DNA extraction [67] and the automated high-throughput DNA protocol [66], which allows DNA extraction to be performed without destroying the specimen. Another integrated

technique available for thrips identification involves piercing the abdominal region of the thrips specimen using a minute pin and processing the extracted gut content for molecular identification prior to the slide mount to preserve the voucher specimen [7]. However, this method requires great skill to keep the specimen intact and save the specimen for slide preparation. Because thrips are soft-bodied minute insects, specimens can be damaged while puncturing the abdomen or during slide preparation. Recently, a new integrated identification technique has been developed for correct identification of thrips using a single specimen. Prior to the DNA extraction of thrips larvae or adults, specimens are subjected to traditional morphological identification using high resolution scanning electron microscopy (SEM) and then gold/palladium sputter coated thrips specimens are processed for polymerase chain reaction assay for molecular identification [14]. Photo-documentation can be easily created for any future reference for the specimen under study. This novel technique has advantages over other integrated methods as it is simple and quick, utilizes fewer specimens for identification, provides high yield of DNA and can be easily mastered by non-experts.

8. Life cycle

Thysanoptera have always been recorded as opportunistic species, as their life history strategies were preadapted from a detritivorous ancestral group developed in a habitat where optimal conditions of survival were brief [68]. Mating does not result in fertilization of all the eggs and unfertilized eggs produce males while fertilized eggs produce females. Sex ratio is in favor of female progeny [16]. The stages of the life cycle of *S. dorsalis* include the egg, first and second instar larva, prepupa, pupa and adult. Gravid females lay eggs inside the plant tissue (above the soil surface) and eggs hatch between 5-8 days depending upon environmental conditions [12, 16]. Larvae and adults tend to gather near the mid-vein or borders of the damaged portion of leaf tissues. Pupae are found in the leaf litter, on the axils of the leaves, and in curled leaves or under the calyx of flowers and fruits. Larval stages complete in 8-10 days, and it takes 2.6-3.3 days to complete the pupal stages. The life span of *S. dorsalis* is considerably influenced by the type of host they are feeding. For example, it takes 11.0 days to become an adult from first instar larva on pepper plants and 13.3 days on squash at 28°C. *S. dorsalis* adults can survive for 15.8 days on eggplant but 13.6 days on tomato plants [25]. They can grow at minimal temperatures as low as 9.7°C and maximum temperatures as high as 33.0°C. Their thermal requirement from egg to egg is 281-degree days and egg to adult is 265-degree days [25]. Populations are multivoltine in temperate regions with up to eight generations per year and 18 generations per year in warm subtropical and tropical areas [69]). In Japan, *S. dorsalis* start egg laying in late March or early April when temperatures are favorable for development (70) and first generation adults can be seen from early May [71]. However, *S. dorsalis* cannot overwinter in regions where temperature remains below -4°C for five or more days [69]. Prolonged rainy seasons do not appear to affect populations much, but the population remains more abundant during prolonged dry conditions than in moist rainy periods.

9. Management strategies

Incursions of *S. dorsalis* are difficult to manage and successful eradication is possible only with early detection and immediate implementation of management practices. Host crops, which develop from seeds such as bean, corn or cotton, must be carefully monitored during the seedling stage of growth because this stage is extremely susceptible to attack by *S. dorsalis* [12]. Symptoms of infestations of *S. dorsalis* must be monitored on their susceptible host plants like roses, pepper, cotton, etc. twice per week and if symptoms appear, then thrips samples should be sent to a reputable laboratory for identification.

9.1. Sampling plan

Appropriate methodology for sampling *S. dorsalis* populations is essential to understand presence and absence of thrips and to determine levels of population abundance at a given time of infestation in a specific host crop. The sampling method has to be economically sound and it should provide information on pest abundance with a minimal number of samples collected. Thus, it is important to determine the within-plant and spatial distribution of the pest in order to select an appropriate sampling unit. For example, melon thrips (*Thrips palmi* Karny) appears on the bottom leaves of most of its vegetable hosts, but on the top leaves of pepper plants. *S. dorsalis* attacks all above ground parts of its hosts, although initiation of infestation can invariably be seen on the young leaves of seedlings and mature plants. As plants grow older, *S. dorsalis* populations may disperse on the entire plant with the abundance on the younger leaves. In a study conducted in St. Vincent [52], *S. dorsalis* developmental stages were observed on all above-ground parts of 'Scotch Bonnet' pepper, *Capsicum chinense* Jacq.' in rainy and dry seasons (Table 2). Mean numbers of *S. dorsalis* adults and larvae were most abundant on the top leaves followed by middle leaves and bottom leaves, flowers and fruits. No significant difference was observed in *S. dorsalis* adults and larvae counts reported on the bottom leaves, flowers and fruits.

In general, insects may have clumped, random or regular distribution in the field and at the initial stage of invasion, insects may appear at a certain location(s) of a crop field depending on environmental factors. These locations may be at the edge of the fields or inside the fields. Known factors that influence such distribution includes wind direction, light intensity, soil fertility, soil moisture, crop vigor and crop nitrogen levels. In several of our studies, *S. dorsalis* displayed various patterns of within-field distribution. The distribution patterns of *S. dorsalis* adults in 2004 and 2005 in a pepper planting were either random or regular in the smaller plots (6, 12 and 24 m²). However, the distribution of adults in the larger plots (48 m²) was aggregated in October 2004 (rainy season), and regular in March 2005 (dry season). Characterizing hot spots (region of aggregation in a field) helps develop an economical sampling methodology and adoption of site selected management strategies using biocontrol agents, lower volumes of insecticides, and effective cultural control practices.

Direct methods of *S. dorsalis* sampling involves counting thrips on any part of a host plant (e.g. leaf, flower and fruit) by using a hand lens, microscope or the naked eye. In this method, the

Location on Pepper plant	Mean number of <i>Scirtothrips dorsalis</i>		
	Adults	Larvae	Total
Field 1 (October 2004, rainy season)			
Top leaf	4.50a	5.50a	10.00a
Middle leaf	1.75b	2.00b	3.75b
Bottom leaf	0.50b	0.75c	1.25c
Flower	0.75b	0.25c	1.00c
Fruit	0.25b	1.00bc	1.25c
Field 2 (March 2005, dry season)			
Top leaf	2.25a	4.25a	6.50a
Middle leaf	1.00ab	2.25ab	3.25b
Bottom leaf	0.25b	0.75bc	1.00c
Flower	0.50b	0.25c	0.75c
Fruit	0.50b	0.75bc	1.25c
Field 3 (March 2005, dry season)			
Top leaf	3.75a	4.00a	7.75a
Middle leaf	1.25b	1.75ab	3.00b
Bottom leaf	0.75b	0.50bc	1.25bc
Flower	0.25b	0.25c	0.50c
Fruit	0.50b	1.00bc	1.50bc

Means within a column for each field followed by the same letter do not differ significantly ($P > 0.05$, Waller-Duncan k ratio procedure).

Table 2. Within plant distribution of *Scirtothrips dorsalis* adults and larvae on 'Scotch Bonnet' pepper plants in three fields in St. Vincent based on samples taken during October 2004 (Field 1), March 2005 (Fields 2 and 3). Source: [52].

part of the plant host sampled may be detached or left intact on the plant. In a beat pan or beat board method, the plant part is tapped against the board to separate *S. dorsalis* adults. More accurately *S. dorsalis* can be sampled by washing plant parts with 70% ethanol or kerosene oil. The contents of the liquid are sieved through a 300 mesh sieve to separate thrips which are then observed using a microscope or hand lens. In an indirect method of *S. dorsalis* sampling, sticky cards of various colors can be placed inside, outside or at the perimeter of the crop field at the level of crop canopy. *S. dorsalis* are attracted to the color and get stuck. Sticky cards can be used from planting to harvest of a crop to monitor thrips advent and abundance during the crop season. Yellow colored sticky cards are commonly used to monitor *S. dorsalis*, but blue, white and green colored cards also attract *S. dorsalis* adults. In a recent study [72] conducted in Taiwan and St. Vincent, three different sticky cards (blue, yellow and white) were evaluated for sampling of *S. dorsalis* and the results suggested that yellow sticky cards could be used efficiently for population detection and monitoring purposes of this pest. In Japan, yellowish-

green, green and yellow sticky boards were found to be effective in attracting *S. dorsalis* adults [73]. Irrespective of colors, sticky cards should be replaced every 7-10 days by a new one.

9.2. Cultural practices

Development of effective management practices for *S. dorsalis* is still in its infancy. The World Vegetable Center has several recommendations which could serve as a basic management practice template for the control of this pest in vegetable production. It involves crop rotation, removal of weeds (which may serve as hosts or virus reservoirs), insecticide rotation and supporting the maximum use of natural enemies including predators and parasites. In some of the plant cultivars resistance to *S. dorsalis* feeding appears to exist. Presence of gallic acid plays a crucial role in resistance to *S. dorsalis* in some varieties of the pepper plant [25]. Recently, researchers at the Mid-Florida Research and Education Center, University of Florida, screened for plant resistant to *S. dorsalis* feeding in 158 different cultivars of pepper and found 14 of these cultivars were resistant to the pest attack. "Brigadier hybrid" and "Trinidad perfume" were among the highly resistant cultivars.

In Japan, synthetic reflective (vinyl) film has been used to protect citrus crops from *S. dorsalis* infestations [74]. In another study, the use of white aqueous solution, i.e. 4% CaCO₃ on mandarin orange trees along with reflective-sheet mulching reported to provide effective suppression of *S. dorsalis* populations [75]. Common cultural practices like vermiwash in addition to the use of vermi-compost and neem (*Azadirachta indica* A. Juss.) cake has also been found effective in regulating *S. dorsalis* attack on pepper [76].

9.3. Chemical control

Chemical control is the primary mode of management of *S. dorsalis* and a wide range of insecticides belonging to different chemical groups is currently used worldwide to control this pest. In south-central Asia, chemical control is conducted using older chemistries including organophosphates such as quinalphos, dimethoate, and phosphamidon as well as the carbamate, carbaryl. In India and Japan monocrotophos, also an organophosphate and the pyrethroid permethrin gave better suppression of this pest (50, 77). Organophosphates (malathion and fenthion) were also found effective against *S. dorsalis* on grapevine [78]. Since their introduction in the Greater Caribbean, there was a lack of information for effective management of this insect using modern insecticides. In recent years, effectiveness of various novel chemistries against *S. dorsalis* has been evaluated and 10 chemical insecticides belonging to seven different modes of action classes (Table 3) have been reported to provide good control of the pest [79, 80]. The rotational use of three or more insecticides from different action classes have been suggested to get prolonged suppression of the pest population [81]. Pyrethroids have not been reported to provide effective control against *S. dorsalis* in the New World and although it causes an instant reduction in pest populations in other parts of the world, it also kills natural controlling agents, ultimately leading to resurgence of pest populations. Various formulations of imidacloprid (neonicotinoid insecticide class), used either as soil drenches or foliar applications provide effective suppression of *S. dorsalis* populations for several days (Table 3) after application of treatments.

Common Name	Trade Name	IRAC Class	Residual Control (days)			
			Foliar		Soil	
			Adult	Larva	Adult	Larva
Abamectin	Agrimek [®] , Avid [®]	6	2	2		
Acephate	Orthene [®]	1B	7	7	7	7
Chlorfenapyr	Pylon [®]	13	7	7	-	-
Dinotefuran	Venom [®] , Safari [®] SG	4A	10	15	0	0
imidacloprid	Marathon [®] , Provado [®] , Admire [®]	4A	15	15	15	15
Novaluron	Pedestal [®] , Ramon [®]	15	7-14	7-14	-	-
Spinosad	Conserve [®] , SpinTor [®]	5	15	15	-	-
Spinetoram	Radiant [®]	5	15	15	15	15
Thiamethoxam	Actara [®] , Platinum [®]	4A	10	15	10	15
Borax + orange oil + detergents	TriCon [®]	8D	10	10	-	-
Beauveria bassiana	Botanigard [®]	Not applicable	3-7	3-7	-	-
Metarhizium anisopliae	Met52 [®]	Not applicable	7	7	-	-

Table 3. Choices of insecticides for rotational use against *Scirtothrips dorsalis* populations. Source: [80]

Management practices from an ecological point-of-view must be environmental friendly but from a growers' viewpoint must be economical, fast acting as well as long lasting. Different chemical insecticides that could satisfy all concerns, like spinetoram and various neonicotinoid insecticides do cause significant reduction in *S. dorsalis* on pepper crops [79]. However, due to their frequent use, insect pests are under intense selection pressure to develop resistance against these insecticides. There are many reports where excessive reliance on insecticides has resulted in resistance development in this pest. In India, resistance in *S. dorsalis* populations has been reported to a range of organochlorine (DDT, BHC and endosulfan), organophosphate (acephate, dimethoate, phosalone, methyl-o-demeton and triazophos) and carbamate (carbaryl) insecticides [82]. Recently, *S. dorsalis* was reported to develop resistance against monocrotophos, acephate, dimethoate, phosalone, carbaryl and triazophos [83]. Thus, in order to prevent or delay development of resistance or minimize the progressive assembly of genes for resistance through selection in the pest against a particular chemistry, it is necessary to rotate insecticides from diverse chemical groups, and explore alternative methods of pest control. Inclusion of effective biorational and biological products in a best management program for *S. dorsalis* can lead to reduced applications of synthetic insecticides. Use of biorational and biocontrol products early in the season will delay the buildup of damaging pest populations on host plants. Furthermore, reduction in the use of harmful insecticides will increase the population of natural biocontrol agents.

9.4. Biological control

Biological control is the active manipulation of beneficial organisms to reduce the pest population below the economic injury level [84]. In this, activity of one species is exploited to reduce adverse effects of another. It is one of the oldest types of pest management strategies. Biological control is employed with the aim of long time pest control by bringing the pest population to non-economic levels. Biological controlling agents are living natural enemies e.g. predators, parasitoid, parasites or pathogens. Various biological controlling agents like minute pirate bugs, *Orius* spp. (Hemiptera: Anthocoridae) and the phytoseiid mites *Neoseiulus cucumeris* and *Amblyseius swirskii* have been reported to provide effective control of *S. dorsalis* on pepper [85, 86]. Adults of *Orius insidiosus* have been observed to feed on all the developmental stages of thrips, and since it is a generalist predator which feeds on aphids, mites, moth eggs and pollen, its population does not decline when there are periodic drops in the thrips population. The biocontrol potential of two phytoseiid mites, *Neoseiulus cucumeris* and *Amblyseius swirskii* evaluated against *S. dorsalis* showed that *A. swirskii* can be a promising tool in managing its population on pepper [86]. In Japan, the predatory mite *Euseius sojaensis* was found to be effective in regulating *S. dorsalis* populations on grapes [87]. Other predatory phytoseiid mites that show promise for biological control include *E. hibisci* and *E. tularensis*. It has been suggested to use two or more natural enemies as a strategy to improve biological control of greenhouse pests [88]. Predators that warrant further study as potential natural enemies of *S. dorsalis* include lacewings (*Chrysoperla* spp.), several mirid bugs, ladybird beetles, and a number of predatory thrips including the black hunter thrips (*Leptothrips mali*), *Franklinothrips* (*Franklinothrips vespiformis*), the six spotted thrips (*Scolothrips sexmaculatus*), and the banded wing thrips (*Aeolothrips* spp.).

The role of entomopathogens like *Beauveria bassiana*, *Metarhizium anisopliae* and *Isaria fumosorosea* in managing field populations of *S. dorsalis* are still under study. *B. bassiana* used with some adjuvants has been reported to control larval populations of *S. dorsalis* for the first few days after application, but soon the population of *S. dorsalis* increases and becomes equivalent to the control plants [80]. In India, significant reduction in *S. dorsalis* populations was reported using entomopathogens *Fusarium semitectum* in pepper fields [89]. However, commercialization and success of this biorational product in different biogeographical regions is still in need of evaluation. Therefore, there is an immense need for developing new strategies to employ best management practices for this serious pest utilizing cultural, chemical and biological control methods.

10. Future prospects

Apart from changing climatic conditions, insect pests are another constraint affecting agricultural production. Insect pests are responsible for loss estimates of 10-20% of main agricultural crops which makes them a major yield limiting factor [90]. To control these pests chemical insecticides are often used by growers on a calendar basis which backfires many times and it leads to a "3R" situation - resistance, resurgence and replacement. To check this situation, it is important to utilize all the resources available in the agroecosystem in a controlled and effective manner. Integrated pest management is an ecosystem-based pest management

strategy which focuses on the longtime control of pests using a combination of techniques, such as cultural control, biological control, habitat manipulation, and use of biotechnological methods. Chemical insecticides are used wisely only after monitoring, under suitable guidelines with the aim to control target pests with no effect on non-target organisms and environment. In the case of *S. dorsalis*, evaluation of chemical insecticides against effective predators like *A. swirskii*, *O. insidiosus* and *E. sojaensis* is needed so that both management systems can exist together.

In the near future, advancement in biological control strategies of *S. dorsalis* could be the use of banker plant systems. Our research group is working in this direction to screen and use different pepper cultivars which could be effectively used as banker plants for the establishment of predatory mites in nurseries and in field conditions. It can effectively solve a number of pest problems in ornamental and vegetable cropping systems including whiteflies, thrips and mites. Banker plant systems also known as open-rearing systems; it is an integrated biological control approach which involves combined aspects of augmentative and conservation biological control and habitat manipulation proposed as an efficient alternative to chemical based pest management techniques. [91, 92, 93]. Success of biological control strategies depends upon the potency of natural enemies against the target pest as well as its adaptability, survival and long-term establishment in the habitat. Installing banker plants in the agroecosystem, ornamental landscape and nurseries can support the establishment of biological control agents by providing suitable ecological infrastructures. The infrastructure can be in the form of a nutrient supplement (nectar/pollen) which is crucial for their survival in the absence of prey, or it can be in the form of a modified microhabitat which can provide protection against adverse abiotic conditions, an insecticide application as well as the hyperpredation/parasitism (secondary enemies) of the agents [94]. The provision of food and shelter reduces mortality of the released biological control agents and may favor their survival, fecundity, longevity and potency to regulate target pests in the habitat thereby supporting the success of biological control strategy.

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Author details

Vivek Kumar^{1,4*}, Garima Kakkar², Cindy L. McKenzie³, Dakshina R. Seal¹ and Lance S. Osborne⁴

*Address all correspondence to: vivekiari@ufl.edu

1 Tropical Research and Education Center, University of Florida, Institute of Food and Agricultural Sciences, Homestead, FL, USA

2 Fort Lauderdale Research and Education Center, University of Florida, Institute of Food and Agricultural Sciences, Davie, FL, USA

3 United States Department of Agriculture, Agricultural Research Services, Fort Pierce, FL, USA

4 Mid-Florida Research and Education Center, University of Florida, Institute of Food and Agricultural Sciences, Apopka, FL, USA

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