PLANT SCIENCE TODAY
Vol x(x): xx–xx
https://doi.org/10.14719/pst.7358

eISSN 2348-1900

REVIEW ARTICLE

An in-depth study of the exotic whiteflies in India's coconut ecosystems: A bibliometric analysis and approaches to their management

S Suriya[1*], G Preetha[2], V Sadhana[2], N Balakrishnan[2], J Sheela[3], G Madhanram[1], Ashish Ajrawat[1] & Showkat Ahmad Sheikh[1]

[1]Division of Entomology, Sher-e- Kashmir University of Agricultural Sciences and Technology of Kashmir, Srinagar 190 025, Jammu and Kashmir, India
[2]Department of Agriculture Entomology, Tamil Nadu Agricultural University, Coimbatore 641 003, Tamil Nadu, India
[3]Department of Plant Pathology, V O Chidambaranar Agricultural College and Research Institute, Tamil Nadu Agricultural University, Killikulam, Vallanadu 628 252, Tamil Nadu, India

*Correspondence email - suriyaento23@gmail.com

Received: 22 January 2025; Accepted: 03 April 2025; Available online: Version 1.0: 18 May 2025

Cite this article: Suriya S, Preetha G, Sadhana V, Balakrishnan N, Sheela J, Madhanram G, Ashish A, Showkat A S. An in-depth study of the exotic whiteflies in India's coconut ecosystems: A bibliometric analysis and approaches to their management . Plant Science Today (Early Access). https:/doi.org/10.14719/pst.7358

Abstract

Coconut trees are constantly threatened by various insect pests, with invasive whiteflies proving particularly harmful. Species such as the rugose spiralling whitefly (*Aleurodicus rugioperculatus*), Bondar's nesting whitefly (*Paraleyrodes bondari*), nesting whitefly (*Paraleyrodes minei*) and palm infesting whitefly (*Aleurotrachelus atratus*) are severely impacting coconut production in tropical and subtropical regions. These whiteflies damage palms by feeding on sap, disrupting nutrient flow and causing yellowing leaves and premature leaf drop. This weakens the palms, making them more vulnerable to diseases. The honeydew they produce encourages the growth of sooty mould, which blocks sunlight and further hampers photosynthesis. These whiteflies are identifiable by their waxy secretions and distinctive spiralling egg-laying patterns. Their ability to affect various hosts, including ornamental plants and crops, complicates control efforts, facilitating their spread. Natural predators, such as parasitoid wasps from the *Encarsia* and *Eretmocerus* genera, help control whitefly populations by targeting eggs and nymphs. Sustainable pest management relies on an integrated pest management (IPM) approach, which combines biological control, regular monitoring, cultural practices like pruning and sanitation and the careful use of chemical insecticides. Adopting a robust IPM strategy is key to controlling whitefly infestations, reducing pest resurgence and ensuring coconut palms' long-term health and productivity while maintaining ecosystem balance and sustainability. A bibliometric analysis using the Bibliometrix R package and VOS viewer (version 1.6.20) has been conducted to assess research trends and identify knowledge gaps.

Keywords: bibliometric analysis; distribution; host plants; invasive whiteflies; management; natural enemies; species validation; symptoms and damage

Introduction

Coconut (*Cocos nucifera* L.), often referred to as the "tree of heaven" or "kalpavriksha," is a vital plantation crop cultivated predominantly in tropical and subtropical regions. Coconuts are nutritionally rich, providing essential minerals, vitamin B, copper, iron, proteins and antioxidants. Coconut water is increasingly popular as a natural functional drink, often marketed as a "sports beverage" due to its hydrating properties (1). Additionally, virgin coconut oil is highly valued as cooking oil, recognized for its medium-chain triglycerides and beneficial antimicrobial and hypolipidemic properties.

Coconuts are grown on approximately 12.26 million hectares worldwide, yielding around 6.67 billion nuts annually, with a productivity rate of 5440 nuts per hectare. The Philippines, Indonesia and India lead in global coconut cultivation, with India growing coconuts on 2.28 million hectares and producing over 20.5 billion nuts annually, achieving a productivity rate of 9,018 nuts/ha (2). Within India, Kerala, Karnataka and Tamil Nadu are the top three states for coconut cultivation (3).

547 insect and mite pests were recorded in coconut palms throughout the year, including 118 exotic species (4). In India, 440 whiteflies from 63 genera species have been identified, indicating a widespread threat to coconut cultivation (5).

Among these pests, the rugose spiralling whitefly (*A. rugioperculatus*), first reported in Belize in 2004, has emerged as a major concern for coconut growers in South India. This pest was identified in several regions, including Kerala and Karnataka, between 2016 and 2019 (6–8). The adults lay creamy golden eggs in a distinctive spiral pattern on the undersides of coconut leaves. Upon hatching, the nymphs feed on plant sap, excreting honeydew that promotes the growth of sooty mold, particularly capnodium, which creates a visible charcoal-black coating on the leaves (9, 10). This fungal growth significantly impairs photosynthesis, leading to decreased yield and nut quality. A survey conducted across Tamil Nadu revealed that the damage caused by *A. rugioperculatus* varied between 40.96 % and 62.86 %, highlighting the urgent need for effective management strategies to protect coconut plantations from this invasive pest (11).

In addition to the rugose spiralling whitefly Bondar's nesting whitefly (*P. bondari*) has emerged as a significant pest in coconut plantations, first reported in Kayamkulam, Kerala, in 2018. This pest has been documented to feed on over 25 host plants posing a serious threat to coconut gardens, particularly in Tamil Nadu (12). The nymphs and adults of *P. bondari* create distinctive nesting chambers made of woolly wax on the undersides of leaflets, where they remain for egg-laying. Another invasive species, the nesting whitefly *P. minei*, was observed in coconut gardens across the Western Ghat coastal regions of Kerala and Karnataka starting in November 2018 (13). This further underscores the growing challenges posed by whitefly infestations in these regions. Additionally, the palm-infesting whitefly, *A. atratus*, was initially reported on ornamental areca palms in Karnataka's Mysore and Mandya districts 2019 (14).

Several natural enemies have been reported to help control whitefly populations. The parasitoid *Encarsia guadeloupae* has been commonly associated with the rugose spiralling whitefly (*A. rugioperculatus*) (7). Additionally, nine different predator species, including coccinellids, chrysopids, cybocephalids and formicids, have been documented preying on *A. rugioperculatus* (15). However, no native parasitoids have been identified again Bondar's nesting whitefly (*Paraleyrodes bondari*) (16). Observations of predators such as coccinellids, psocids and chrysopids on the invasive *P. minei* highlight the potential for biological control (13). Establishing these natural enemies and microbial biocontrol agents like *Isaria fumosorosea* are effective in controlling whiteflies by infecting and killing them in coconut gardens, which could favour the bio-suppression of whiteflies. While numerous insecticides are commercially available for whitefly control, their indiscriminate use can cause multiple issues. These include insect resistance, pest resurgence, secondary pest outbreaks, disruption of natural enemy complexes, biodiversity loss and environmental pollution (17). Moreover, applying insecticides to the undersides of tall fronds is logistically challenging. In response, the Government of India and ICAR declared a 'pesticide holiday' for coconut whiteflies, shifting focus toward evaluating the efficacy of potential biocontrol agents.

This article examines invasive whiteflies' occurrence, distribution and impact on coconut, covering symptoms, damage and population dynamics. It discusses species identification through morphological and molecular methods, host plants and natural enemies. Effective management strategies are also highlighted, including biological, cultural and chemical control. By emphasizing integrated pest management approaches, the goal is to mitigate the impact of these invasive whitefly species on coconut cultivation while promoting sustainable agricultural practices.

Occurrence and distribution

Rugose spiralling whitefly, *Aleurodicus rugioperculatus*

The rugose spiraling whitefly is a significant exotic pest that has severely impacted coconut-growing regions in India. Initially identified by Martin in Belize in 2004, it spread to other parts of Central and North America, including Mexico, Guatemala and Florida, where it caused damage to various plants, including gumbo limbo trees and coconut fronds (18, 19).

In India, the whitefly was first documented in Tamil Nadu in July-August 2016, in Pollachi, Coimbatore district (7). Its presence was soon reported in the coastal areas of Andhra Pradesh by late 2016 (20). A comprehensive review highlighted its widespread distribution across states such as Karnataka, Kerala, Tamil Nadu, Maharashtra, Gujarat, Himachal Pradesh and Sikkim, noting heavy infestations and the resulting secondary sooty mold infections affecting both coconut palms and surrounding horticultural crops (21). The pest continued to spread, with reports documenting significant infestations in Mandouri, West Bengal, in June 2019, marking its first appearance in that region (22). By 2020, reports also emerged of *A. rugioperculatus* in the Saurashtra region of Gujarat, further underscoring its increasing threat to coconut cultivation across India (23) Bondar's nesting whitefly, *Paraleyrodes bondar* was first described in 1971 by Peracchi, on citrus species in Brazil. As a native of the Neotropical region, this pest has since expanded its range and has been reported in several locations, including Belize, Honduras, Puerto Rico, Madeira, Comoros, Mauritius, Taiwan, Hawaii and Florida (USA) (19). In Florida, *P. bondari* was first recorded in 2011 when specimens were collected from a ficus hedge in Lee County. This marked the species' identification outside its native range. In 2018, a study reported a severe outbreak of *Paraleyrodes bondari* on cassava in Uganda (24). In India, *P. bondari* was first reported in 2019, explicitly infesting coconut trees in Kerala (25). Following this, researchers provided the first evidence of the species' invasion and establishment in the Andaman and Nicobar Islands (16).

In Tamil Nadu, the whitefly was identified in coconut plantations in Namakkal District, with previously reported findings (26). Their research noted that *P. bondari* was not limited to coconut; it was also found on a variety of other crops, including bhendi (*Abelmoschus esculentus*), chillies (*Capsicum annuum*), white poplar (*Populus alba*), banana (*Musa* spp.), guava (*Psidium guajava*) and the ornamental plant *Duranta erecta* in Coimbatore.

Nesting whitefly, *Paraleyrodes minei*

Paraleyrodes minei was first described in 1989 on *Citrus aurantium* leaves collected from the coastal region of Syria.

Paraleyrodes minei (nesting whitefly) in Syria, despite being native to the Neotropical region, is likely due to human-mediated introduction, mainly through the international trade of live plants (EPPO global database). It was reported in California in 1984, first noticed near San Diego and the southern coast. Over the years, *P. minei* has been documented in various locations, including Belize, Guatemala, Mexico, Puerto Rico, Bermuda, Texas, Lebanon, Morocco, Spain, Turkey and Benin (27). Research highlighted the invasive nature of *P. minei* as it spread to regions like Hong Kong and Hainan (28). The pest has shown a significant global spread, reaching the Western Palaearctic region, including countries like Iran, Israel and Turkey. It was first reported in Greece in June 2015 on sweet orange leaves in Platanias, Chania (29).

In the Andaman and Nicobar Islands of India, *P. minei* has been recorded on various host plants, including Punnai (*Calophyllum inophyllum*), Noni (*Morinda citrifolia*) and guava (*Psidium guajava*) (30). The pest was later observed along the western Ghat coastal regions of Kerala and Karnataka in November 2018 (13). In Tamil Nadu, a survey of whitefly species affecting horticultural crops revealed the presence of *P. minei* on coconut (*Cocos nucifera*) and banana (*Musa* spp.) in Coimbatore (26). All four whitefly species (*Paraleyrodes minei, Aleurodicus rugioperculatus, Paraleyrodes bondari and Aleurotrachelus atratus*) coexist in coconut ecosystems in Tamil Nadu and contribute to crop damage. However, specific quantitative data on their individual or combined damage impact is currently unavailable. Their simultaneous presence poses a significant challenge for pest management, as they collectively affect plant vigor, promote sooty mold formation and reduce coconut yield.

Palm infesting whitefly, *Aleurotrachelus atratus*

Palm infesting whitefly was first described from coconut in Brazil in 1922, quickly spread to various regions, including Antigua, the Bahamas, Barbados, Bermuda, Colombia, Venezuela and Florida (USA) (31). *Aleurotrachelus atratus* has been reported to cause significant damage to coconut palms in the Comoro Islands (32) and has also been observed in the southwestern Indian Ocean islands (33). *Aleurotrachelus atratus* infests palm leaves, causing yellowing, wilting and premature leaf drop due to sap-sucking. The honeydew excreted by the whiteflies promotes sooty mold growth, which reduces photosynthesis and weakens the plant. Severe infestations lead to reduced yield in commercial palms and aesthetic damage in ornamental species. In England, large populations of *A. atratus* infesting African oil palm (*Elaeis guineensis*) were documented and it is believed that the species arrived in 2001 through coconut palms imported from the Netherlands (34).

In India, *A. atratus* was first reported in coconut plantations of Karnataka and on ornamental areca palm (*Dypsis lutescens*) in the Mandya and Mysore districts (14). Subsequently, its presence was confirmed on coconut (*Cocos nucifera*) in the Dharmapuri and Krishnagiri districts of Tamil Nadu (35). Further reports from Kannur, Kerala, highlight its continued spread and potential impact on palm cultivation in the country (36).

Taxonomy and dynamics of invasive whiteflies in India

The whitefly fauna of India is characterized by a significant diversity, comprising a total of 66 genera. Of these, 64 belong to the subfamily Aleyrodinae (Westwood), which includes several economically important genera. Notable among these are *Bemisia* (38), *Trialeurodes* (39) and *Aleurothrixus* (37). These genera are recognized for their roles as pests in agriculture, particularly due to their capacity to transmit plant viruses and cause substantial crop damage. In addition to Aleyrodinae, the subfamily Aleurodicinae (Quaintance & Baker) accommodates four genera: *Aleurodicus* (Douglas), *Aleuronudus* (Hempel), *Metaleurodicus* (Back) and *Palealeurodicus* (Martin). The diversity within these subfamilies underscores the ecological complexity of whiteflies in India.

Among the notable invasive species in India are whiteflies from the genera *Aleurodicus*, including *A. dispersus* and *A. rugioperculatus*, along with two species from the genus *Paraleyrodes*: *P. minei* and *P. bondari*. The genus *Paraleyrodes* includes seventeen species, which contribute to the challenges of managing whitefly populations. This genus features species such as *P. citricolus, P. triangulae, P. goyabae*, *P. proximus*, *P. persea, P. pseudonaranjae*, *P. singularis*, *P. urichii* *P. citri*, *P. ancora, P. crateraformans*, *P. cervus, P. pulverans*, *P. perplexus* and *P. naranjae*, as documented previously (39, 27). The invasive whitefly complex has been taxonomically classified from kingdom to genus as follows: Animalia, Arthropoda, Insecta, Hemiptera, Sternorrhyncha, Aleyrodoidea, Aleyrodidae, Aleurodicinae and *Paraleyrodes*. This systematic classification highlights the hierarchical structure used in identifying and categorizing whiteflies, reflecting their biological relationships and evolutionary history.

Key characters of taxonomical identification

The puparium of *A. rugioperculatus* was identifiable by its broadly cordate vasiform opening, which is complemented by a spinulose operculum on the ventral-basal side and a distinctly rugose texture on the dorsal surface. It includes a pair of fine setae at the ventro-median position, while the lingula head extends beyond the vasiform opening, showcasing a finely spinulose texture with an acute tip. Four additional setae are located near the apex. This puparium can be clearly differentiated from *A. dispersus*, another species in India, by small compound pores on abdominal segments VII and VIII, a distinctly rugose operculum and an unusually narrowly acute lingual apex (40). The study highlighted distinct puparial characteristics that differentiate the rugose spiralling whitefly from the spiralling whitefly (27). The rugose spiralling whitefly exhibits a reticulated dorsal cuticle, compound pores on abdominal segments VII and VIII, surface corrugations on the operculum and an acutely shaped lingula. In contrast, the spiralling whitefly has a smooth cuticle, lacks compound pores on abdominal segments VII and VIII and possesses an oval-shaped lingula apex (6).

The puparial features of *P. bondari* consist of a larger cephalic pore and four abdominal compound pores, surrounded by an outer ring of ovoid cellular facets that resemble stylized flower petals. The last four abdominal compound pores are complemented by simple discoidal pores and 2–3 additional discoidal pores paired with two smaller abdominal pores, each about half the size. This arrangement results in 7–8 facets resembling flower petals. The male genitalia are notable for their distinctive aedeagus, which features a single dorsal and ventral horn at the apex and a pair of apicolateral processes.

The puparial features of *P. minei* include a larger cephalic pore, four abdominal compound pores and an outer ring of ovoid cellular facets that create a stylized flower petal appearance. The last four abdominal compound pores are accompanied by simple discoidal pores, 2–3 additional discoidal pores and two reduced abdominal pores, each about half the size of the larger ones. Together, these structures form 8–9 facets that mimic flower petals. The tongue-like lingula extends beyond the posterior margin of the vasiform orifice and has two pairs of apical setae, while the operculum partially covers both the lingula and the vasiform orifice. The male claspers feature a robust nail and the aedeagus has a distinctive cockhead-shaped apex, which includes three short appendices on the upper and posterior surfaces, alongside two lengthy, thin appendices that project downward beneath the shorter anterior appendix. The distinguishing characters of Bondar's Nesting Whitefly and Nesting Whitefly were similarly outlined (40, 41). Both studies emphasize specific taxonomical traits that differentiate these species, providing valuable insights for accurate identification.

The puparium of *A. atratus* is elongated and oval-shaped, with a smooth dark cuticle. The marginal teeth are distinct and separated, featuring converging either subtruncate or rounded tips, each with serrated edges. Notably, no first abdominal or mesothoracic setae are present. The metathoracic setae extend beyond the second abdominal segment, while the eighth abdominal setae are longer than the vasiform orifice. The caudal setae are very long and located on tubercles. The submarginal area is characterized by rows of flat, elongated granules of similar size and the tip of the lingula is rounded.

Additionally, each puparium displays a distinct pair of diagnostic sub-marginal longitudinal cephalothoracic folds that extend into the abdomen. The same distinguishing characters were were also noticed (40). Their descriptions provide key morphological traits for accurately identifying and differentiating *A. atratus* from other related species.

Species validation by morphological characterization

Distinct characteristics mark the life stages of the rugose spiraling whitefly (RSW). The adult female lays eggs in a spiral pattern on palm surfaces, while the nymphs exhibit subtle projections arranged in a tail-like formation. *A. rugioperculatus* nymphs undergo five instars, with the first instar being a crawler that hatches from the egg and seeks a feeding site using its needle-like mouthparts to extract plant sap. The convex puparium features a triangular rugose operculum and a pointed lingula. Adult RSW are robust, displaying a white colouration adorned with greyish-brown mottling on their wings (Fig. 1).

Adult rugose spiralling whiteflies have long, pincer-like appendages at the tip of their abdomen. The nymphs produce slender, waxy filaments and dense cottony wax, with their colouration ranging from light to golden yellow (27, 42). RSW eggs are yellowish and elliptical, covered with white flocculent wax. During the immature stages, nymphs produce abundant wax filaments, including long, glassy, crystal-like rods and tufts of fluffy wax. Adult RSW are slightly larger than common whiteflies and are characterized by two light brown bars running along their wings (14, 23).

Paraleyrodes bondari, commonly known as Bondar's Nesting Whitefly, is characterized by its unique appearance and behavior. The adult whiteflies exhibit distinctive"-shaped oblique greyish stripes on their wings, which serve as a visual identifier for this species. Combined with their small size, these markings make them easily distinguishable from other whiteflies (Fig. 2).

One of the most notable features of *P. bondari* is its nesting behaviour. The females produce woolly wax nests on palm leaves' abaxial (underside) surface. These nests consist of a dense layer of waxy material, protecting the eggs and nymphs developing within (25). One of the keys identifying features of *P. minei* is its wing structure, characterized by a median and forked radial vein, which distinguishes it from other whiteflies. Additionally, the spine -like tarsal paronychium on the legs enhances its ability to adhere to host plants, an adaptation crucial for its survival. While *P. minei* shares similarities with *P. bondari*, it notably lacks the oblique grey stripes characteristic of the latter species.

Furthermore, *P. minei* constructs loosely woven fuzzy wax nests, which serve as a protective environment for its eggs and nymphs. This nesting behaviour is a significant aspect of its life cycle, reflecting its ecological adaptations. Egg-laying behaviour is also noteworthy; *P. minei* produces cream-coloured egg clusters with short stalks that transition to a pinkish hue during eclosion, indicating the developmental stages of the embryos within. The nymphs are described as flat and creamy-yellow, adorned with fibreglass threads on their dorsum, providing additional protection against predators and environmental stressors.

Regarding sexual dimorphism, male *P. minei* individuals exhibit a smoky grey colouration, complemented by smoky grey wings. Their antennae are uniquely orange-tinged and whip-like, consisting of three segments that play a role in sensory perception. In contrast, female whiteflies are predominantly white, featuring a grey blotch in the terminal area of their wings. Their antennae are four-segmented with a swollen second segment, further differentiating them from males (Fig. 3). The morphological traits of *Paraleyrodes minei* have been consistently documented, highlighting their importance in the identification and classification of this species within the Aleyrodidae family (43, 44).

The eggs and nymphs of *A. atratus* exhibit distinct behavioural and morphological traits, primarily congregating on the underside of coconut fronds. They are densely covered with flocculent white waxy material,

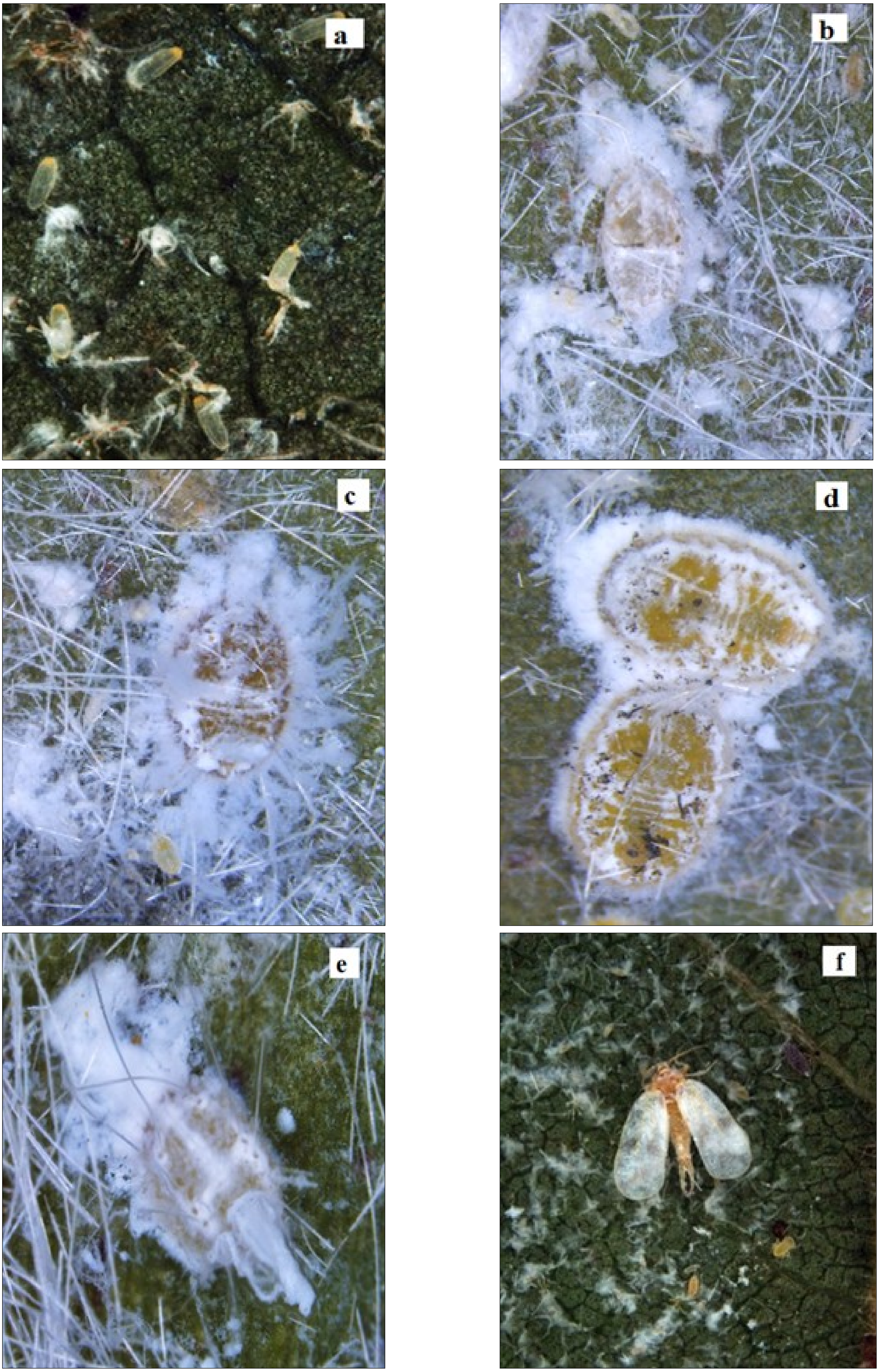

Fig. 1. Developmental stages of *A. rugioperculatus*: a. eggs are laid in a spiral pattern; b. 1st nymphal instar; c. 2nd nymphal instar; d. 3rd nymphal instar; e. 4th nymphal instar; f. adult.

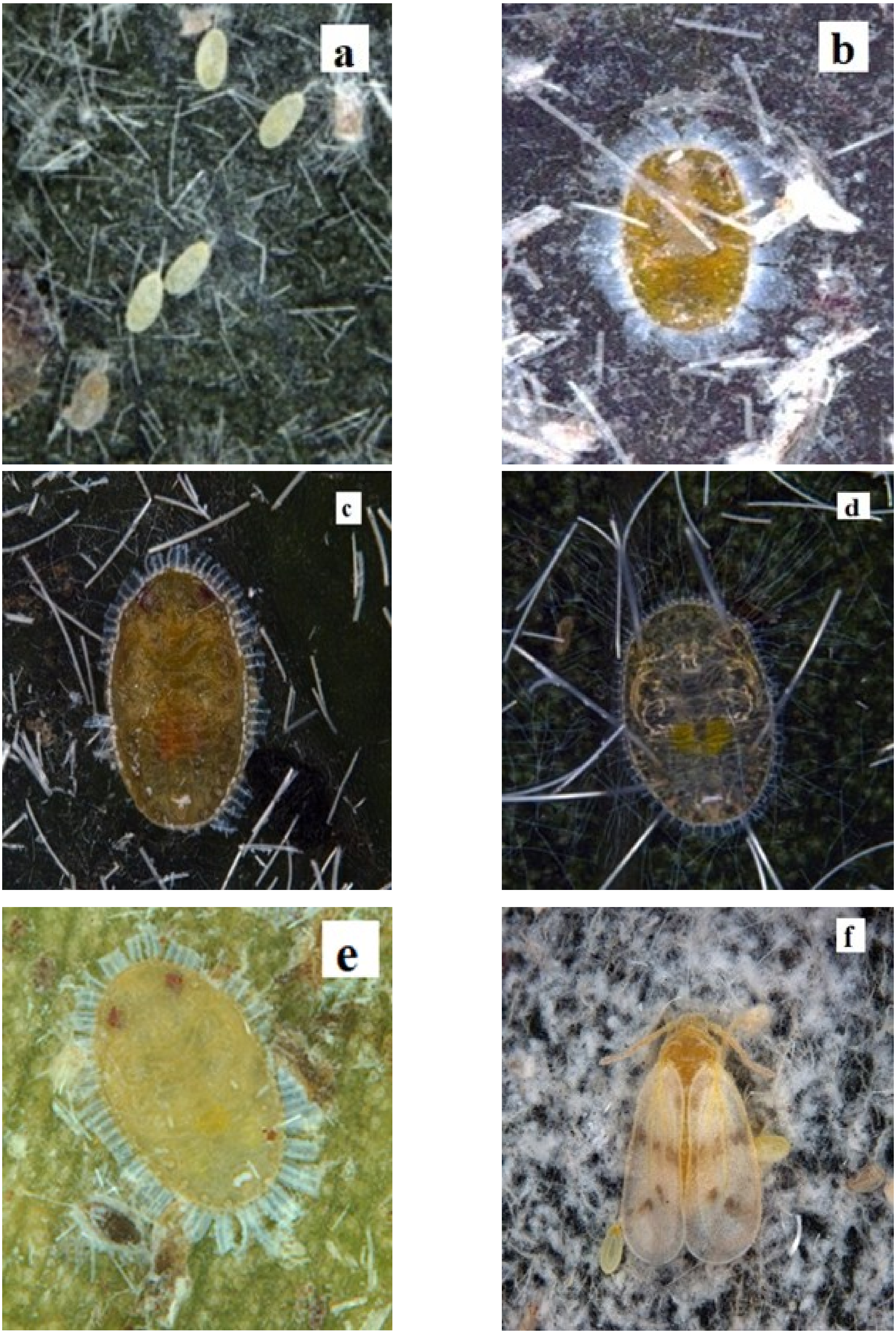

Fig. 2. *P. bondari* life stages: a. eggs; b-e. nymphs; f. male and female adult.

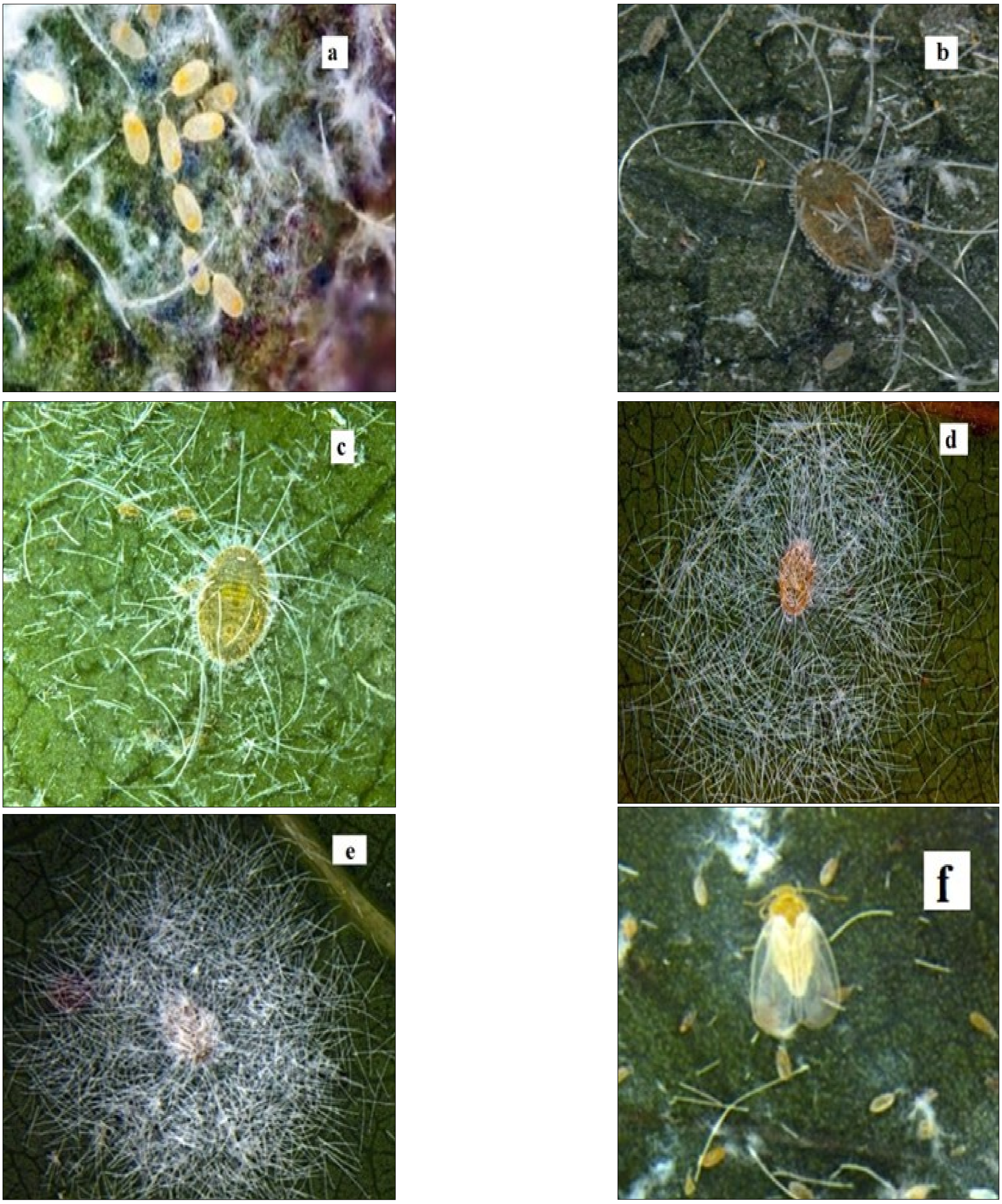

Fig. 3. *P. minei* life stages: a. eggs; b-e. nymphs; f. adult.

which serves as a protective barrier over the black pupae beneath. Adult *A. atratus* are more significant than both *Paraleyrodes bondari* and *Paraleyrodes minei*, making them easily distinguishable in the field. A key feature of their wings is the absence of any wavy markings, which sets them apart from other species (Fig. 4). These described features are essential for the identification and classification of *A. atratus*, aiding in the understanding of its ecological interactions and potential impact on coconut crops (14).

Biology and morphometric analysis

The detailed study of the biology and morphometric analysis of the invasive whitefly complex on coconut is summarized in Table 1. This table provides a comprehensive overview of key measurements, life cycle stages and developmental durations for various species of whiteflies affecting coconut cultivation. It includes information on egg sizes, nymphal instar dimensions, pupal characteristics, adult measurements and the duration of each life cycle stage. The data highlights the differences and similarities among the invasive species.

The life cycle of *A. rugioperculatus* spans approximately 59 days, with the egg stage lasting about 6.9 days, nymphs about 19.57 days and pupae around 10.9 days. Females lay eggs in a spiral pattern on leaf undersides and the nymphal stage lasts about 27.7 days before transitioning to adulthood, which has an average lifespan of 20.5 days (11, 45).

Morphometric measurements of rugose spiralling whiteflies revealed that male adults averaged 2.63 mm in

Fig. 4. *A. atratus* life stages: a. eggs; b-c. nymphs; d. adult.

Table 1. Morphometric measurements and life stage durations of whitefly species reared on coconut

Life stages	Mean (± SE) (mm)		Mean (± SE) Duration (Days)
	Length	Breadth	
Aleurodicus rugioperculatus			
Eggs	0.26 ± 0.02	0.14 ± 0.02	6.54 ± 0.43
1st nymphal instar	0.37 ± 0.04	0.25 ± 0.02	5.51 ± 0.72
2nd nymphal instar	0.56 ± 0.04	0.27 ± 0.01	5.23 ± 0.48
3rd nymphal instar	0.87 ± 0.08	0.36 ± 0.01	8.12 ± 0.72
4th nymphal instar	1.11 ± 0.09	0.73 ± 0.08	10.1 ± 0.75
Adult	2.52 ± 0.08	1.71 ± 0.15	21.8 ± 2.54
Total life cycle	-	-	62.6 ± 0.64
Paraleyrodes bondari			
Eggs	0.18 ± 0.05	0.09 ± 0.02	5.24 ± 0.21
1st nymphal instar	0.26 ± 0.02	0.15 ± 0.01	3.18 ± 0.17
2nd nymphal instar	0.39 ± 0.04	0.28 ± 0.03	6.95 ± 0.23
3rd nymphal instar	0.48 ± 0.02	0.36 ± 0.02	6.72 ± 0.30
4th nymphal instar	0.63 ± 0.04	0.43 ± 0.07	4.37 ± 0.28
Adult	1.10 ± 0.01	0.72 ± 0.06	15.5 ± 0.37
Total life cycle	-	-	61.51 ± 0.92
Paraleyrodes minei			
Eggs	0.34 ± 0.02	0.22 ± 0.38	5.9 ± 0.20
1st nymphal instar	0.49 ± 0.02	0.54 ± 0.29	5.4 ± 0.34
2nd nymphal instar	0.57 ± 0.04	0.73 ± 0.59	3.5 ± 0.47
3rd nymphal instar	0.64 ± 0.07	0.87 ± 0.45	6.7 ± 0.28
4th nymphal instar	0.84 ± 0.03	0.93 ± 0.33	5.1 ± 0.44
Adult	1.11 ± 0.07	1.71 ± 0.15	11.2 ± 0.76
Total life cycle	-	-	44.6 ± 0.87
Aleurotrachelus atratus			
Eggs	0.12 ± 0.03	0.07 ± 0.01	5.4 ± 0.10
1st nymphal instar	0.29 ± 0.01	0.11 ± 0.07	11.7 ± 0.28
2nd nymphal instar	0.37 ± 0.03	0.21 ± 0.01	8.5 ± 0.04
3rd nymphal instar	0.52 ± 0.07	0.33 ± 0.03	7.3 ± 0.01
4th nymphal instar	1.03 ± 0.08	1.56 ± 0.19	5.9 ± 0.17
Adult	1.95 ± 0.05	1.20 ± 0.04	10.0 ± 0.01
Total life cycle	-	-	48.8 ± 0.61

* Mean length and width were calculated from 15 observations for each life stage. Mean duration was recorded from 15 samples for each life stage at the beginning

length and 2.12 mm in width, while females measured 3.15 mm in length and 2.17 mm in width. The measurements across different life stages were as follows: eggs measured 0.34 mm in length, first instar nymphs 0.41 mm, second instar nymphs 0.73 mm and the oval-shaped puparium 1.59 mm (46). Slightly different measurements were reported, with eggs averaging 0.24 mm in length. The first instars measured 0.35 mm in length and 0.24 mm in width, the second instars 0.58 mm by 0.27 mm, the third instars 0.83 mm by 0.38 mm and the fourth instars 1.08 mm by 0.70 mm. adult males averaged 2.27 mm in length and 1.30 mm in width, while females measured 2.59 mm in length and 1.71 mm in width (47).

Adults of *Paraleyrodes bondari* measure approximately 1.11 mm in body length (13). Morphometric measurements recorded eggs at 0.15 mm in length and 0.08 mm in width, nymphs at 0.46 mm by 0.36 mm, pupae at 0.59 mm by 0.41 mm and adults at 1.09 mm in length and 0.73 mm in width (47). Eggs measured 0.21 mm by 0.10 mm, with first instar nymphs measuring 0.29 mm by 0.16 mm, second instars 0.52 mm by 0.35 mm, third instars 0.59 mm by 0.41 mm and fourth instars 0.77 mm by 0.52 mm. Adults averaged 1.10 mm in length and 0.56 mm in width (48).

The life cycle of *P. bondari* lasts between 20 and 26 days on coconut, with four nymphal stages totalling 15.4 days. The first instar develops in about 3.2 days, the second and third instars take roughly 7 days and the fourth instar lasts around 4.8 days. Adults have flattened wings that extend over the body, a pale-yellow colour with a light wax coating and a lifespan of approximately 16.7 days. Overall, the life cycle can extend to 35 to 40 days on cotton (49).

The nesting whitefly, *Paraleyrodes minei*, was a tiny insect whose morphological and developmental characteristics have been the focus of extensive research, shedding light on its life cycle and growth stages. The eggs of *P. minei* are notably small, measuring approximately 0.368 ± 0.03 mm in length. After a period of incubation lasting around 6 to 7 days, these eggs hatch into first instar nymphs, also known as crawlers, which measure about 0.527 ± 0.04 mm. As the nymphs progress through their life cycle, they undergo several instars, each marked by significant changes in size. The second and third instar nymphs grow to about 0.739 ± 0.08 mm, while the fourth instar reaches a length of 0.867 ± 0.03 mm. Upon reaching adulthood, *P. minei* measures approximately 1.106 ± 0.09 mm in body length, positioning it as a relatively small member of the whitefly family (13).

The life cycle of *P. minei* consists of distinct stages, beginning with the egg stage, where eggs are laid on the undersides of leaves. Following the egg stage, the nymphs undergo four instars, each varying in duration: the first instar lasts about 3 days, the second around 5 to 7 days, the third approximately 4 to 5 days and the fourth about 4 to 5 days. In total, the nymphal stages typically span between 15 to 20 days, influenced by environmental conditions and the health of the host plant. The entire life cycle of *P. minei*, from egg to adult, can take approximately 30 to 45 days under optimal conditions. While specific survival temperature ranges for *P. minei* are not extensively documented, studies on related whitefly species suggest that temperatures below 20 °C or above 30 °C can reduce survival rates.

Molecular characterization

Phylogenetic tree analysis

The analysis involved 56 nucleotide sequences of whitefly complexes specifically targeting the COI (Cytochrome c Oxidase I) gene region. These sequences were obtained from the NCBI database and represent whitefly populations from various countries: China, Korea, Kenya, Florida (USA), Uganda, Mexico, Israel, Taiwan, Colombia and India.

The whitefly populations were classified into three major clades based on the analysis of their COI sequences. This study involved annotating and manually curating *whitefly* mtCOI sequences using the DNA MAN program before submission to GenBank. Sequence similarity was assessed through comparisons with GenBank reference sequences and phylogenetic analysis included available mtCOI sequences, incorporating appropriate out-group species. Sequence refinement was conducted in BioEdit, with multiple sequence alignments performed using Clustal W in MEGA-X software. A phylogenetic dendrogram was constructed using the Neighbor-Joining method with 1000 bootstrap replicates in MEGA-X. The first clade comprises two species: *Paraleyrodes bondari* and *Paraleyrodes minei*. The second clade was represented by *Aleurotrachelus atratus* from the same study. The third clade includes two species, *Aleurodicus rugioperculatus* and *Aleurodicus disperses*. The sequences were likely aligned and analyzed to determine genetic relationships and phylogenetic clustering based on the COI gene. The division into clades suggests distinct genetic groups within the whitefly populations studied. Various studies provide a comprehensive geographical representation of *whitefly* complexes, shedding light on their genetic diversity and evolutionary relationships. *Aleurodicus rugioperculatus* was amplified using a universal COX-I primer, resulting in a 658 bp sequence (7). Partial mitochondrial COX-I sequences of 675 bp and 621 bp were obtained from *Paraleyrodes bondari* (25,16). Genetic analysis revealed high nucleotide similarity among *P. bondari* populations across different regions (25). Kenyan sequences closely matched Indian isolates from guava (GenBank: MW488201) and coconut (GenBank: MW488198), with 99.84 % and 100 % similarity, respectively. A 632 bp sequence from Uganda (GenBank: MH178372) showed 100 % similarity with a Florida isolate (GenBank: KP032215), indicating minimal genetic divergence. Likewise, Indian sequences (GenBank: MK333262) exhibited 100 % similarity with those from Uganda and Florida, reinforcing genetic homogeneity across continents.

In India, *P. bondari* populations attacking the caterpillar tree (MW488193) and infesting oil palm (MW704277) are closely related. This suggests a genetic similarity between these two populations of *P. bondari* within India. In Uganda, the *P. bondari* population infesting cassava (MH178372) shows genetic similarity to the Indian population of *P. bondari* infesting sugarcane (MZ026894). This indicates a genetic relationship between these populations despite the geographical distance between Uganda and India.

Manilkara zapota (MW741558) and coconut (MW750441) infesting populations of *P. minei* in India show a high similarity of 99 %. This suggests that these two populations share a very recent common ancestry despite infesting different host plants. Citrus (MW741558) infesting populations of *P. minei* in India are closely related to the Indian coconut (MW488186) infesting population. This indicates genetic similarity between *P. minei* populations infesting citrus and coconut in India. The remaining populations of *P. minei* were scattered into single clades, suggesting more genetic diversity compared to the highly similar populations.

All the populations of *A. atratus* from coconut and palm trees in India formed a single cluster. This clustering suggests that these populations are genetically very similar to each other. Populations of *A. rugioperculatus* infesting coconut (sequences OK037183 and OK042272) in India were found to be 99.9 % identical to each other based on comparing the data available in NCBI. This high similarity indicates that these populations are closely related genetically, likely suggesting a recent common ancestry or ongoing gene flow between them. Indian sequences of *A. rugioperculatus* infesting maize (OP020879) were closely related to sequences from cotton (OK376251) infesting populations. This suggests a genetic affinity between the populations infesting different host plants (maize and cotton) in India.

The population of *A. dispersus* from Taiwan infesting maize (AY764031) was genetically close to the population infesting cassava (MF149998) in India. This genetic closeness suggests that despite the geographical and host plant differences, these populations of *A. dispersus* share a significant genetic similarity. In summary, the study analyzed 56 COI sequences of whitefly complexes from diverse geographic locations, identifying three major clades corresponding to different species or groups within the whitefly populations. This approach helps understand the genetic diversity and relationships among whitefly populations globally. (Fig. 5).

Molecular identification of *A. rugioperculatus* utilized the cytochrome oxidase I gene (658 bp), registered as GenBank Acc. No. KY209909 (7). The same gene was sequenced for multiple accession numbers (MK883218, MK883219, MK883220, MK926750, MK926751) (50). *A. rugioperculatus* was documented in West Bengal and recorded under Accession No. OP024192 (51). For *P. bondari*, a 632 nt mtCOI sequence matched 100 % with samples from Uganda (GenBank Acc. No. KP032215) (23). Its presence was confirmed with sequence ON739183 (52), while additional molecular analysis was conducted under Accession No. OP024193 (51). *P. minei* was characterized through DNA extraction and COI gene amplification (13). *A. atratus* was listed under GenBank Accession No. MT422351 (53), with further identification corresponding to OQ844114 (54).

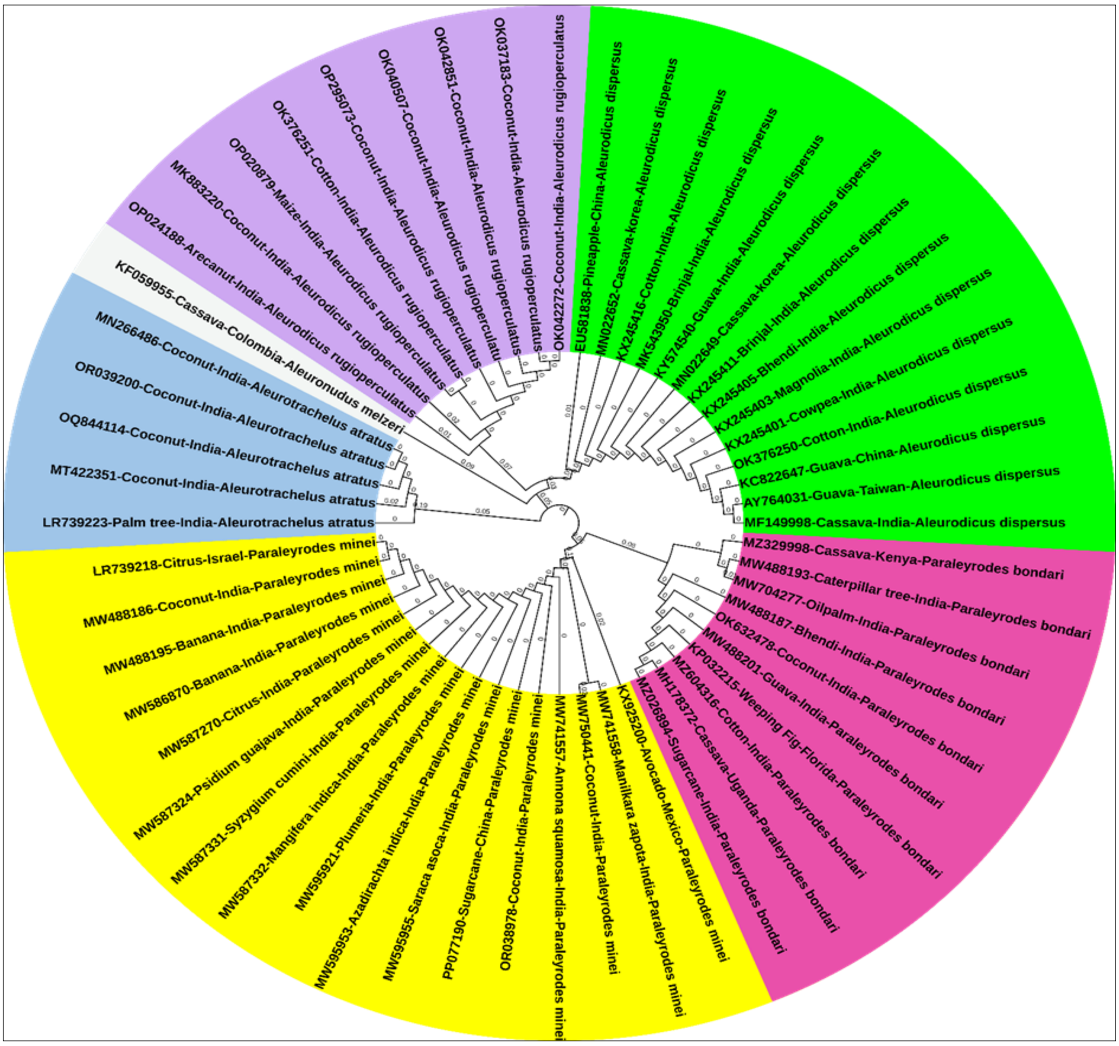

Fig. 5. Phylogeny tree analysis of invasive whiteflies in coconut.

Trends in research on invasive whiteflies: A bibliometric analysis

Search engine and data

The bibliometric mapping approach is data-driven, relying extensively on computer algorithms and visualization techniques. It visually represents a topic by illustrating the relationships between key terms in the field. One of the most widely used software tools for this purpose is VOS Viewer (developed by the Centre for Science and Technology Studies, 2018), which generates co-occurrence maps using keywords (both author-provided and indexed). The maps typically guide the analysis, but expertise remains essential for accurately interpreting them. Bibliometric mapping also produces clusters of keywords (whitefly or *Alerurodicus Paraleyrodes* or *Aleurotrachelus* and invasive or invasion) which require expert review. The expert must analyze each cluster and make sense of the data. This approach can reduce bias, as the expert is not limited by their own area of specialization (55). However, a drawback of this type of study is that some valuable publications may be overlooked due to the vast number of documents analyzed. Finally, it is worth noting that bibliometric studies do not involve human or animal subjects; thus, ethical approval from an Institutional Review Board is not required. This literature review employed a quantitative approach, utilizing two methods: performance analysis (examining publications based on authors, countries and institutions) and science mapping (using bibliometric software) (56). The analysis of publications was carried out through keyword-based studies.

A bibliometric analysis was conducted using the Dimensions AI database, focusing on 446 research articles related to invasive whiteflies and four key species: *Aleurodicus rugioperculatus*, *Paraleyrodes bondari*, *Paraleyrodes minei* and *Aleurotrachelus atratus*. Initially, searches using these species' names as keywords within titles, abstracts, keywords and full-text papers yielded 446 documents, which were analyzed for country-wise contributions. For analyses of annual scientific production, average citations per year, most relevant sources, most relevant authors, globally cited documents and country-wise scientific production, the species names were used as keywords to search within titles, abstracts and keywords, resulting in 169 papers. The Bibliometrix R package facilitated a comprehensive exploration of research trends, authorship patterns and thematic developments, while VOS viewer (version 1.6.20) was employed to enhance and refine the analysis further. These four species of whiteflies were recently introduced in India and limited research papers are available on the topic. As a result, we used a minimal number of sources for the bibliometric analysis.

Results and Discussion

As shown in Fig. 6, the bibliometric coupling of countries revealed India and the United States as the central research hubs, with strong collaborative links to countries such as the United Kingdom, China and Brazil. The network visualization also highlights the emergence of recent contributions from countries like Indonesia and Bangladesh, underscoring a growing interest in these mite species in diverse regions. These findings provide insights into the geographical distribution and intensity of research efforts on these species (Fig. 7). presents author-wise information on whitefly research from 1994 to 2024. The data reveals that Selvaraj has authored the highest number of documents, followed by Sundararaj, Hedge and Vinayaka. The coloured connecting lines represent collaborations between these authors.

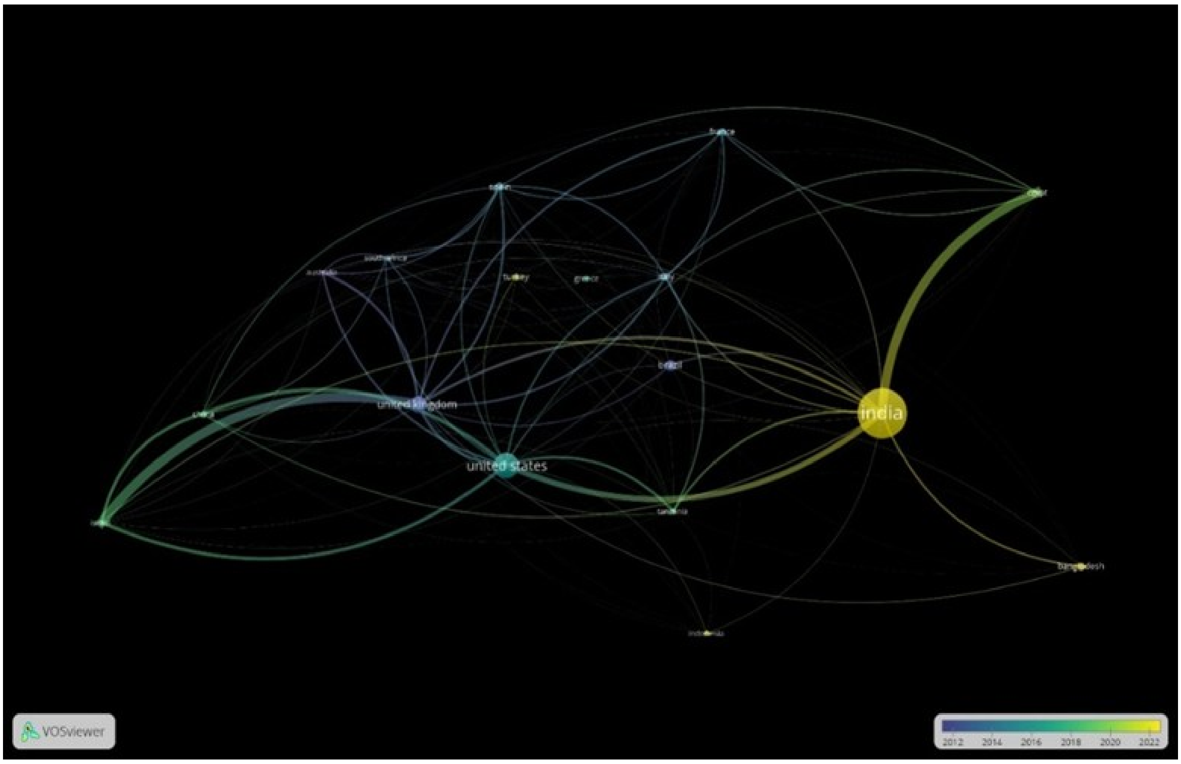

Fig. 6. VOS viewer overlay visualization for invasive whiteflies country wise from 2012 to 2024.

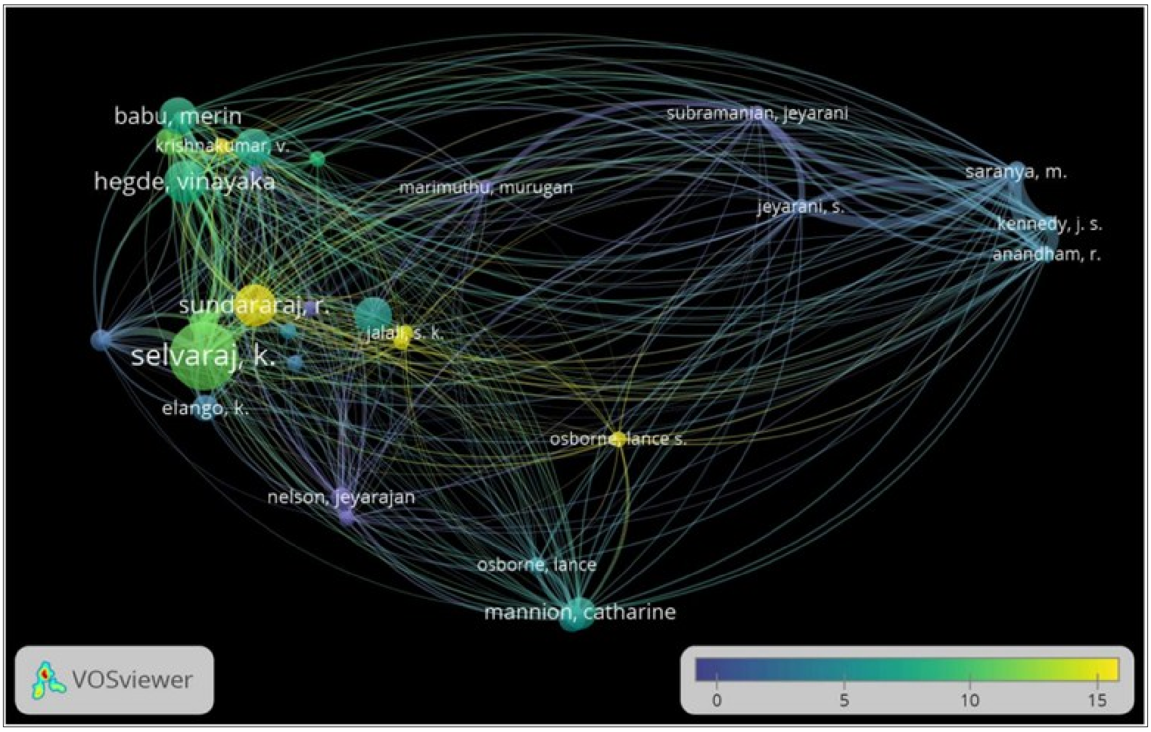

Fig. 7. VOS viewer overlay visualization for author wise whitefly documents information from 1994 to 2024.

Table 4 and Fig. 8 present data on annual scientific production, indicating that whitefly research began in 1996. The peak in publications occurred in 2023 with 31 articles, followed by 30 in 2022 and 23 in 2021. Table 5 and Fig. 9 provide data on average citations per year, showing that 2023 recorded the highest number of citations, 31, surpassing all other years. Table 6 and Fig. 10 highlight the most relevant sources of whitefly research across the top 15 countries. The findings reveal that the Indian Journal of Entomology and Phytoparasitica published the highest number of articles (12), followed by Research Square (9 articles), the International Journal of Tropical Insect Science (7 articles) and the Journal of Biological Control (7 articles). Table 7 and Fig. 11 present the top 10 most relevant authors who have contributed to research on invasive whiteflies. The data shows that Selvaraj K published the highest number of articles (15), followed by Josephrajkumar A (11 articles) and Hedge (8 articles). Table 8 and Fig. 12 highlight the top 10 most cited papers and authors in the field of invasive whiteflies, including globally cited documents. The results show that Sundararaj R (2017) received the highest number of citations (54), followed by Francis AW (2016) with 38 citations. Table 9 and Fig. 13 present country-wise scientific production, showing that India leads in research output, followed by the USA and the UK.

Symptoms of damage and yield loss by invasive whiteflies

The nymphs of whiteflies feed on the sap from the underside of leaves, causing honeydew to exude onto the upper surfaces of leaves. This honeydew promotes the growth of the *Capnodium* fungus, resulting in a black, sooty appearance on affected plants and also negatively impacts photosynthesis, leading to physiological disorders, leaf deformities, immature nut drop and stunted growth, ultimately reducing nut production.

Surveys conducted in Tamil Nadu, Kerala Andhra

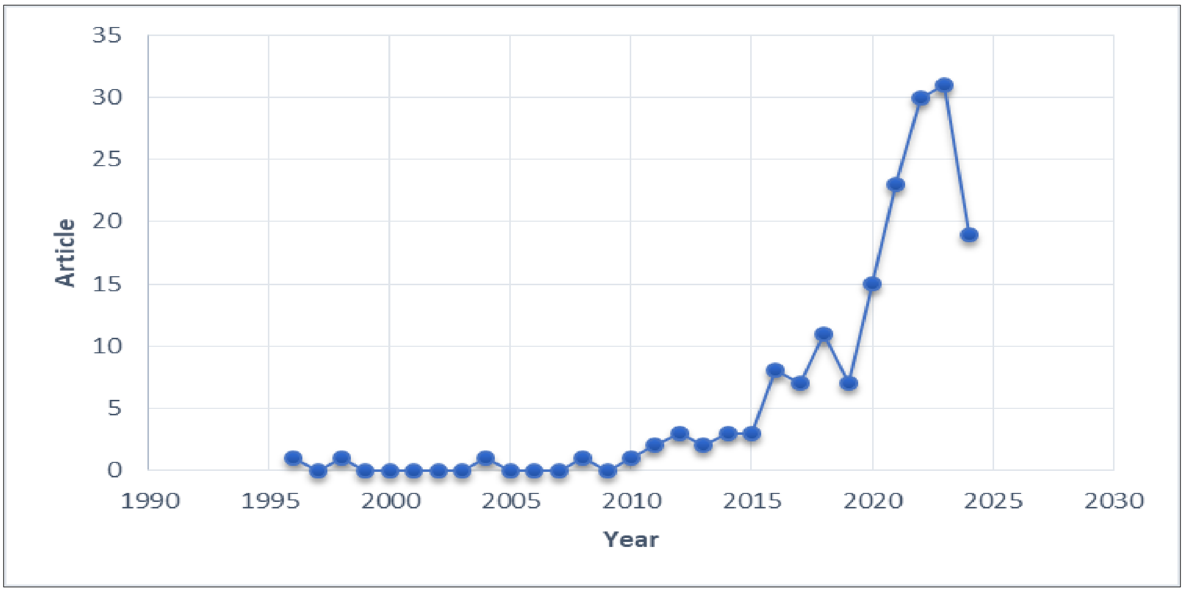

Fig. 8. Annual scientific production of invasive whiteflies.

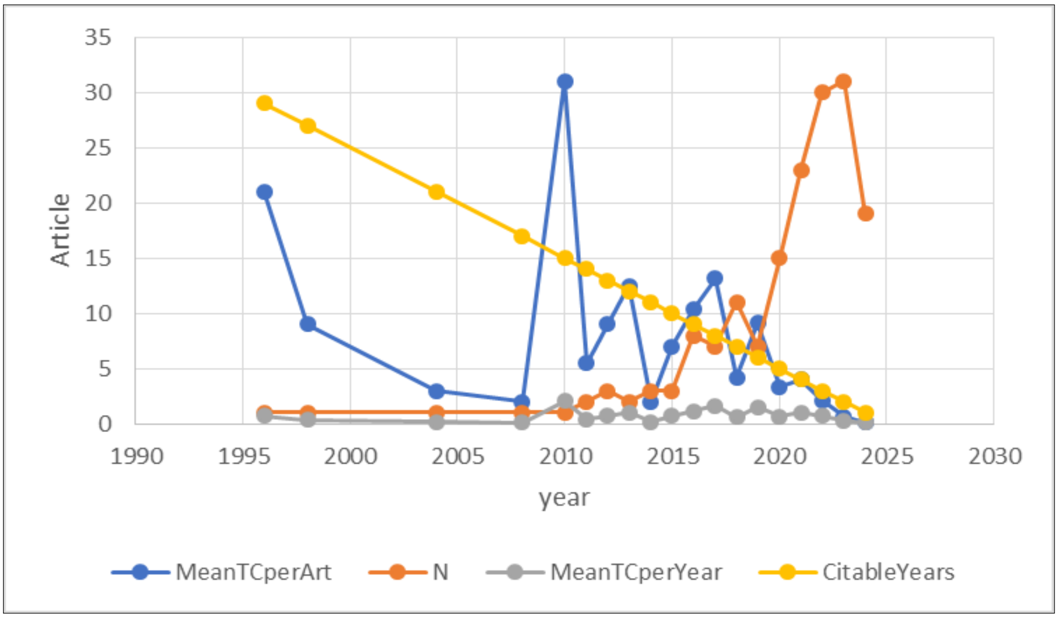

Fig. 9. Average citations per year of invasive whiteflies.

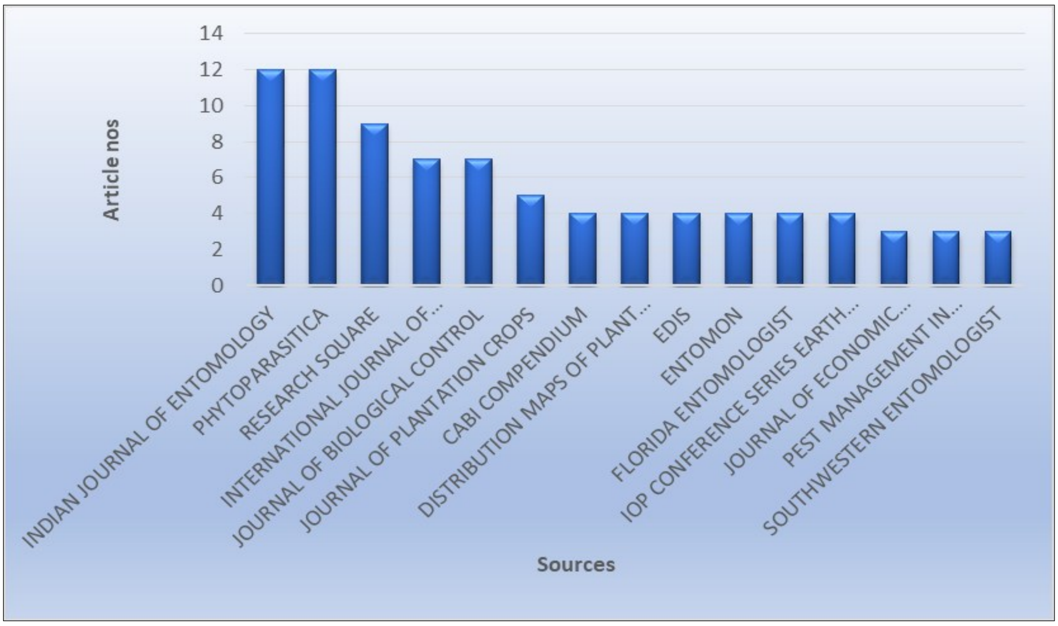

Fig. 10. Most relevant sources of invasive whiteflies.

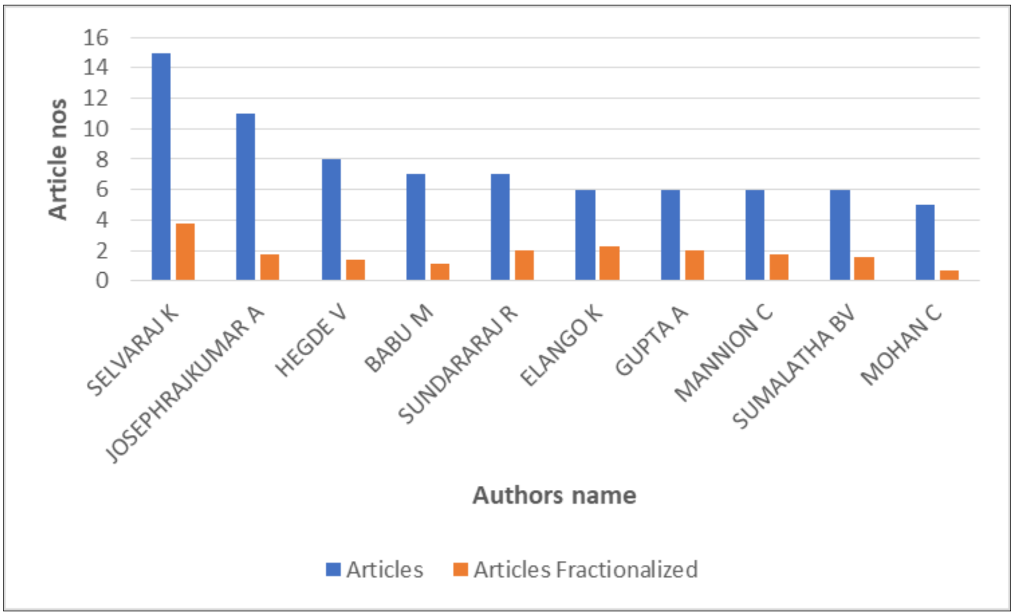

Fig. 11. Most relevant authors of invasive whiteflies.

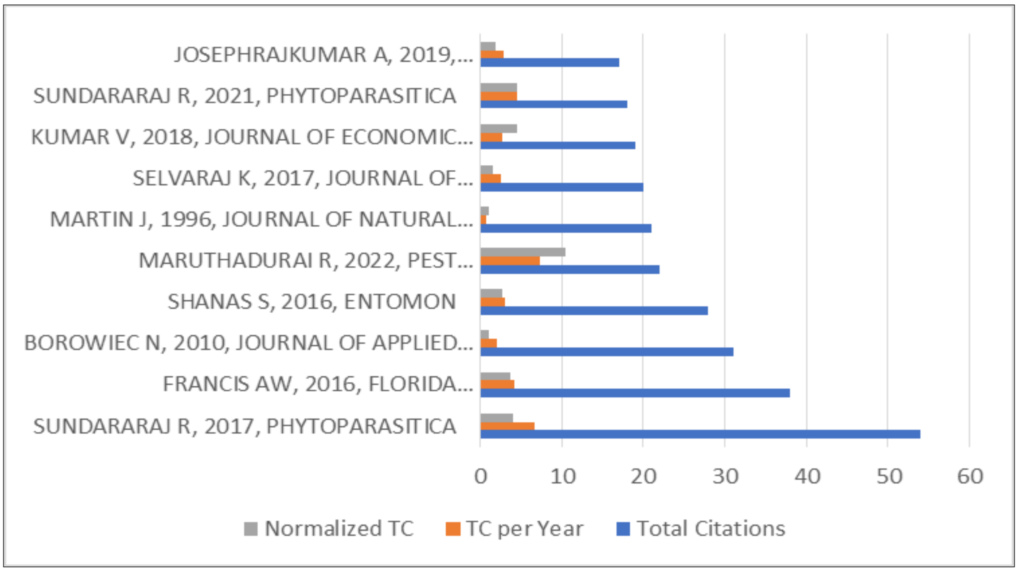

Fig. 12. Globally cited documents of invasive whiteflies.

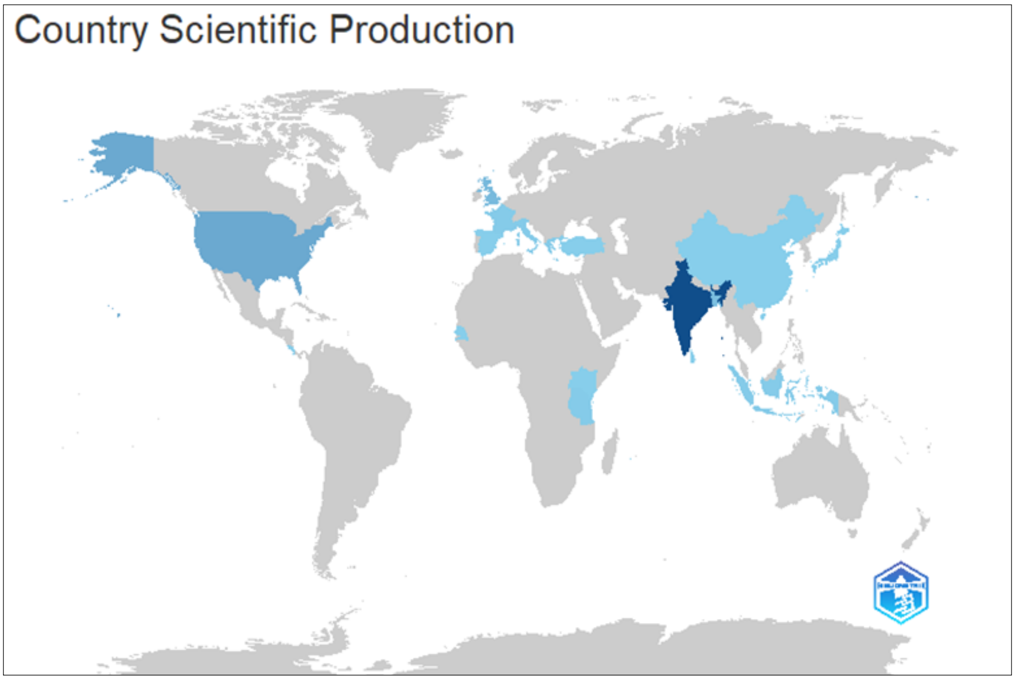

Fig. 13. Country-wise scientific production of invasive whiteflies.

Pradesh and Karnataka showed significant variation in infestation severity, with coconut palms experiencing 40-60 % damage and banana leaves sustaining 25-40 % damage (7). Infested trees were easily identifiable due to the thick sooty mould covering, which caused frond drooping and wilting symptoms (10, 21). *A. rugioperculatus* has caused significant damage to coconut crops, particularly in coastal regions like Mangalore and Udupi, where infestations led to losses of 20-35 % (7). A study in Jashore in 2019 found infestation rates between 46.66 % and 68.33 %, with Magura Sadar Upazila experiencing the highest levels (68.33 %) (57). State assessments indicated infestation levels ranging from 20 % to 100 %, with native coconut varieties showing higher susceptibility (85.7 %) compared to dwarf varieties (81.2 %) (58). Seasonal trends revealed peaks in April-May and September-October, influenced by temperature fluctuations (59). Varying yield losses were reported in hybrid palms, emphasizing the economic impact of these infestations (60).

In Tamil Nadu, Coimbatore district had the highest infestation (62.86 %) from August 2017 to February 2019, with subsequent surveys revealing even higher rates in 2019 -20, particularly in Tirunelveli (75.4 %) and Kanyakumari (75.8 %) (10, 44). Surveys in Ratnagiri, Maharashtra, noted peak infestations during the rainy season (61).

Nests of *P. minei* are small and waxy white, formed from the secretions of females and filamentous wax rods from late instar nymphs (29). These whiteflies establish 3 to 30 nests on coconut leaflets, promoting intense sooty mold growth that adversely affects plant health (25). The damage caused by *A. atratus* is intensified by the sooty mold developing on honeydew excreted by the whiteflies, significantly impacting coconut palm growth and yield (32). Research in Inhambane province revealed that *A. atratus* was the most prevalent whitefly species, constituting 75 % of infestations, while *Bemisia tabaci* was the least common (4.3 %). Notably, a 1 % increase in whitefly severity was associated with a yield loss of approximately 77.13 kg/ha (62). Additionally, *A. atratus* was linked to a staggering 55 %

economic yield loss for coconut growers (14).

Host plants

Coconut trees face year-round pest threats, intensified by climate change, insecticide resistance and habitat destruction. The decline of natural enemies due to pesticide overuse and the spread of invasive pests through trade further worsen infestations. Sustainable integrated pest management (IPM) is crucial for effective control and long-term coconut production. Key pests include the rugose spiralling whitefly (*Aleurodicus rugioperculatus*), Bondar's nesting whitefly (*Paraleyrodes bondari*), the nesting whitefly (*Paraleyrodes minei*) and the palm infesting whitefly (*Aleurotrachelus atratus*), all of which have caused significant damage in major coconut-growing regions of India since their introductions in 2016, 2018 and 2019. In this study, the author documented several host plants affected by *P. bondari* for the first time. These host plants include red ginger, snake, jasmine and egg fruit. This finding enhances our understanding of the ecological impact of *P. bondari* and highlights its potential threat not only to coconut plantations but also to other ornamental crops. These invasive whiteflies are polyphagous, allowing them to feed on various host plants. While they primarily target coconut trees and other broad-leaved plants in their native habitats, their host range has expanded to include multiple economic and horticultural crops and weed species. For detailed information on host plants affected by these invasive whiteflies, refer to Table 2.

Population dynamics

In Coimbatore, Tamil Nadu, it was found that dwarf and hybrid coconut palms aged 4-6 years were highly susceptible to RSW, with the lower leaves exhibiting higher infestation rates ranging from 18 to 37 nymphs/cm^2 (63). Nymph populations were reported to range from 26.02 to 27.72 per leaflet, while puparium populations varied between 33.12 and 33.64 (55). In Chhattisgarh, RSW

Table 2. Host plants of invasive whiteflies

1. Rugose spiralling whitefly, *Aleurodicus rugioperculatus*

Common name	Scientific name	Family	Reference
Broadleaf arrowhead	*Sagittaria latifolia* Willd.	Alismataceae	
Brazilian peppertree	*Schinus terebinthifolia* G. Radd.	Anacardiaceae	
Red mombin	*Spondias purpurea* L.	Anacardiaceae	
Madagascar Periwinkle	*Catharanthus roseus* (L.) G. Don	Apocynaceae	
Norfolk Island pine	*Araucaria heterophylla* (Salisb.) Franco.	Araucariaceae	
Christmas Palm	*Adonidia merrillii* (Becc.) Becc.	Arecaceae	
Thatch Palm	*Coccothrinax* spp. Sarg.	Arecaceae	
Coconut	*Cocos nucifera* L.	Arecaceae	
Princess Palm	*Dictyosperma album* (Bory) Scheff.	Arecaceae	
Triangle palm	*Dypsis decaryi* (Jum.) Beentje & J. Dransf.	Arecaceae	
Areca palm	*Dypsis lutescens*(Wendland) Beentje & Dransfield.	Arecaceae	
Palmiste marron	*Hyophorbe verschaffeltii*H.A. Wendl.	Arecaceae	
Ivory Cane Palm	*Pinanga coronate* (Blume.)	Arecaceae	
Miniature Date Palm	*Phoenix roebelenii* O Brien	Arecaceae	
Foxtail palm	*Wodyetia bifurcate*A.K. Irvine.	Arecaceae	
Cabbage palmetto	*Sabal palmetto* (Walt.) Lodd.	Arecaceae	(42)
Montgomery Palm	*Veitchia arecina* Becc.	Arecaceae	
Mexican fan palm	*Washingtonia robusta* H. Wendl.	Arecaceae	
Rose	*Rosa* spp. L.	Rosaceae	
Field mustard	*Brassica rapa* L.	Brassicaceae	
Gumbo limbo	*Bursera simaruba*(L.) Sarg.	Burseraceae	
Cocoplum	*Chrysobalanus icaco*L.	Chrysobalanaceae	
Button Mangrove	*Conocarpus erectus* var. sericeus Fors ex DC	Combretaceae	
Copperleaf	*Acalypha wilkesiana* Mull. Arg.	Euphorbiaceae	
Florida Keys blackbead	*Pithecellobium keyense* Britton ex Britton & Rose.	Fabaceae	
live oak	*Quercus virginiana* Mill.	Fagaceae	
Ti Plant	*Cordyline fruticose* (L.) A.Chev.	Liliaceae	
Earleaf greenbrier	*Smilax auriculate* Walter.	Liliaceae	
Indian tulip tree	*Thespesia populnea* (L.) Sol	Malvaceae	
Florida strangler fig	*Ficus aurea* Nutt.	Moraceae	
Common fig	*Ficus carica* L.	Moraceae	
Natal wild banana	*Strelitzia Nicolai*Regel & Korn.	Streliziaceae	

Crane flower	*Strelitzia reginae* Banks.	Streliziaceae	
Southern wax myrtle	*Myrica cerifera*L.	Myricaceae	
White stopper	*Eugenia axillaris* (Sw.) Willd.	Myrtaceae	
Surinam Cherry	*Eugenia uniflora*L.	Myrtaceae	
Twinberry	*Myrcianthes fragrans* (Sw.McVaugh	Myrtaceae	
Malabar plum	*Syzygium cumini* (L.) Skeels.	Myrtaceae	
Lawn orchid	*Zeuxine strateumatica* (L.) Schltr.	Orchidaceae	(42, 95)
Satinleaf	*Chrysophyllum oliviforme* L.	Sapotaceae	
Mimusops	*Manilkara roxburghiana* (Wight) Dubard	Sapotaceae	
False mastic	*Sideroxylon foetidissimum* Jacq.	Sapotaceae	
Willow bustic	*Sideroxylon salicifolium* (L.) Lam.	Sapotaceae	
Paradise-tree	*Simarouba glauca* DC.	Simaroubaceae	
Virginia creeper	*Parthenocissus quinquefolia* (L.) Planch.	Vitaceae	
Cassava	*Monihot esculenta* Crantz.	Euphorbiaceae	
Arecanut	*Areca catechu* L.	Arecaceae	
Nutmeg	*Myristica fragrans*Houtt.	Myristicaceae	
Neem	*Azadirachtaindica*A. Juss.	Meliaceae	
Congress grass	*Parthenium hysterophorus* L.	Asteraceae	
Banana	*Musa* spp. L.	Musaceae	
Mango	*Mangifera indica* L.	Anacardiaceae	(8)
Sapota	*Manilkara zapota* (L.) P.Royen	Sapotaceae	
Mexican Petunia	*Monihot esculenta* Crantz.	Euphorbiaceae	
Kenanga	*Areca catechu* L.	Arecaceae	
Cassava	*Myristica fragrans*Houtt.	Myristicaceae	
Arecanut	*Azadirachtaindica*A. Juss.	Meliaceae	
Nutmeg	*Parthenium hysterophorus* L.	Asteraceae	
Mexican Petunia	*Ruellia simplex* C. Wright.	Acanthaceae	
Kenanga	*Cananga odorata* (Lam.) Hook. f. & Thomson.	Annonaceae	
Philodendron	*Philodendron selloum*	Araceae	
Seashore palm	*Allagoptera arenaria* Kuntze	Arecaceae	
Alexandra Palm	Archontophoenix alexandrae (F.Muell.) H. Wendl. & Drude	Arecaceae	
Bangalow palm	*Archontophoenix cunninghamiana* (H. Wendl.) H. Wendl.& Drude	Arecaceae	
Parlour palm	*Chamaedorea* spp.	Arecaceae	
Solitaire palm	*Ptychosperma elegans* (R.Br.) Blume.	Arecaceae	
Florida Royal Palm	*Roystonea regia* (Kunth) O.F. Cook.	Arecaceae	
Queen palm	*Syagrus romanzoffianam* (Cham.) Glassman	Arecaceae	
Malabar spinach	*Basella alba* L.	Basellaceae	
Java cotton	*Ceiba* spp.	Bombacaceae	
Bandanna of the everglades	*Canna flaccida* Salisb.	Cannaceae	
Antilles beauty leaf	*Calophyllum antillanum* Britton.	Calophyllaceae	
Oriental persimmon	*Calophyllum Brasiliense* Cambess.	Calophyllaceae	
Machete ice-cream-bean	*Diospyros kaki* Thunb.	Ebenaceae	
Subabul	*Inga* spp. Mill.	Fabaceae	
False tamarind	*Leucana leucocephala* (Lam.) de Wit.	Fabaceae	
Horseflesh Mahogany	*Lysiloma latisiliquum* (L.) Benth	Fabaceae	
Florida Keys blackbead	*Lysiloma sabicu* Benth.	Fabaceae	
Indian beech	*Pithecellobium keyense* Britton ex Britton & Rose.	Fabaceae	
Laurel oak	*Pongamia pinnata* (L.) Pierre.	Fabaceae	(95)
Scrub hickory	*Quercus laurifolia* Michx.	Fagaceae	
Bay laurel	*Carya floridana* Sarg.	Juglandaceae	
Lancewood Tree	*Laurus nobilis* L.	Lauraceae	
Pride of India	*Ocotea coriacea* (Sw.) Britton	Lauraceae	
Hazel Sterculia	*Lagerstroemia speciosa (*L.) Pers.	Lythraceae	
Weeping fig	*Sterculia foetida* L.	Malvaceae	
Spanish stopper	*Ficus benjamina* L.	Moraceae	
Strawberry guava	*Eugenia foetida* Pers.	Myrtaceae	
Rose Apple	*Psidium cattleyanumSabine*	Myrtaceae	
	Syzygium jambos (L.) Alston	Myrtaceae	
Wild pepper	*Bougainvillea sp*	Nyctaginaceae	
Pigeon Plum	*Piper sarmentosum* Roxb. ex W. Hunter	Piperaceae	
Sea Grape	*Coccoloba diversifolia* Jacq.	Polygonaceae	
Kaffir Lime	*Coccoloba uvifera (*L.) L.	Polygonaceae	
Biscayne Prickly Ash	*Citrus hystrix* DC.	Rutaceae	
Longan	*Zanthoxylum coriaceum*	Rutaceae	
Quenepa	*Dimocarpus longan* Lour.	Sapindaceae	
Traveller's Palm	*Melicoccus bijugatus* Jacq.	Sapindaceae	
Possum grape	*Ravenala madagascariensis* Sonn.	Strelitziaceae	
West Indian Holly	*Cissus verticillata* (L.) Nicolson & C.E.Jarvis	Vitaceae	
Muscadine Grape	*Leea guineensis*G. Don	Vitaceae	
Shell ginger	*Vitis rotundifolia* Michx	Vitaceae	
	Alpinia zerumbet (Pers.) B. L. Burtt and R. M. Sm.	Zingiberaceae	

Guava	*Psidium guajava*L.	Myrtaceae	(42, 95)
Cashew	*Anacardium occidentale*L.	Anacardiaceae	(14)
Water apple	*Syzygium aqueum* (Burm.f.) Alston	Myrtaceae	
Indian shot	*Canna indica* L.	Cannaceae	
False bird of paradise	*Heliconia* spp. L.	Heliconiaceae	
Tomato	*Solanum lycopersicum* L.	Solanaceae	(26)
Maize	*Zea mays* L.	Poaceae	(14, 96)
Indian almond	*Terminalia catappa*L.	Combretaceae	(42)
Jackfruit	*Arthocarpus heterophyllus*Lam.	Moraceae	(95)
Bottle palm	*Hyophorbe lagenicaulis* (L.H. Bailey) H. E. Moore.	Arecaceae	
Nutmeg	*Myristica fragrans* Houtt.	Myristicaceae	(21)
Saptree	*Garcinia* spp. (L.)	Clusiaceae	
Taro	*Colacasia* spp. Schott.	Araceae	
Moringa	*Moringa oleifera* Lam.	Moringaceae	(42)
Custard apple	*Annona* sppL.	Annonaceae	

densities ranged from 14.2 to 30.6 per cm^2, with the highest density observed in Gautami Ganga (64). An average of 21.05 spirals per leaflet was found in East Godavari, while West Godavari recorded a significantly higher average of 124.09, indicating substantial damage across leaflets, petioles and nuts (65). Minimal infestations were noted during the rainy season, with a significant increase in summer, peaking at 161 nymphs per leaf in October (46).

Infestation levels in Karnataka varied, with lower numbers recorded in the northern transition zone and significantly higher levels in the southern zone (66). Peak whitefly populations reached 34.1 in April (59), with fluctuations ranging from 4.24 to 102.87 adults per frond, peaking in mid-April (65). In Veppankulam, 28.6 adults per leaflet were documented (67), while egg spirals and nymph counts ranged from 9.36 to 23.41 and 13.41 to 34.17, respectively (68). Surveys in Tamil Nadu revealed adult populations between 29.50 and 34.60 per leaflet, with the highest counts observed in Kanyakumari (47). In coastal districts of Bangladesh, the highest RSW abundance was recorded in Khulna, with 23 egg spirals and 34 nymphs per leaflet, whereas Patuakhali showed lower counts (68).

Population Dynamics Research has highlighted the population trends of *P. bondari* in various regions. *P. bondari* was reported to have a nymphal population of approximately 8.04 nymphs per 30 cm leaflet in coconut, indicating a concerning level of infestation (16). More recently, a significant increase in population was observed, with 31.1 adults per leaflet recorded in Dharmapuri (67). Biweekly surveys conducted in southern Tamil Nadu from December 2020 to August 2021 revealed notable variations in whitefly populations. Kanyakumari recorded the highest nymph population at 35.31 per leaflet and the highest adult population at 34.84 per leaflet. In contrast, Tenkasi had the lowest nymph count at 22.79, while Thoothukudi reported the lowest adult count at 24.19 (47).

Natural enemies

Controlling invasive whiteflies through the use of natural enemies, such as predatory insects and parasitoids, offers a sustainable and environmentally friendly method for managing their populations. Details on natural enemies of invasive whiteflies can be found in Table 3. Whiteflies are known for their rapid reproduction and damaging effects on crops, but their numbers can be effectively reduced by leveraging natural predators. Beneficial insects like ladybugs and lacewings actively seek out and consume whiteflies at various life stages, significantly diminishing their populations and mitigating plant damage. Additionally, parasitoids, such as certain wasp species, enhance biological control by laying eggs inside whitefly nymphs. As the parasitoid larvae develop, they feed on the host, leading to its death and preventing further reproduction. This disrupts the whitefly life cycle while minimizing the reliance on chemical pesticides, which can harm non-target organisms and the environment.

Management

Invasive whiteflies pose a significant threat to coconut palms by sucking sap and depositing honeydew, which encourages the growth of sooty mould. Effective management of these pests requires an integrated approach, combining cultural, biological, chemical and physical control methods. Cultural practices interrupting the whitefly life cycle and reducing favourable conditions are essential. Crop rotation with non-host plants can help break the pest's life cycle and limit population growth (69). Regular pruning of infested fronds and removal of dead leaves can also effectively lower whitefly populations (70).

Additionally, high-pressure water sprays can dislodge whiteflies from coconut leaves, especially during the early stages of infestation (61). Removing weeds that act as alternative hosts for whiteflies can significantly reduce the risk of infestation Research in various cropping systems has identified several weed species that support whitefly populations, including *Abutilon indic*um (Indian mallow), *Chrozophora rottleri* (suryavarti), *Solanum nigrum* (black nightshade) and *Hibiscus ficulneus* (white wild musk mallow) (ISWS). Removing these weeds disrupts the whitefly life cycle and reduces infestation risks.

On the other hand, certain non-host plants can help deter whiteflies by acting as repellents or trap crops. Examples include marigold (*Tagetes* spp.), neem (*Azadirachta indica*), lemongrass (*Cymbopogon* spp.) and basil (*Ocimum basilicum*), which have been observed to repel whiteflies or reduce their populations in agricultural settings (71). Trap cropping with marigolds or sunflowers, which attract whiteflies, can divert them away from the main crops (72).

Table 3. Natural enemies of invasive whiteflies

S. No.	Predators	Family & Order	References
	Predators of invasive whiteflies		
	1) Rugose spiralling whitefly, *Aleurodicus rugioperculatus*		
	Nephaspis oculate Blatchley.	Coccinellidae: Coleoptera	
	Azya orbigera orbigera Mulsant	Coccinellidae: Coleoptera	
	*Chilocorus cacti*L.	Coccinellidae: Coleoptera	
	Cryptolaemus montrouzieri Mulsant.	Coccinellidae: Coleoptera	
	*Delphastus pallidus*LeConte.	Coccinellidae: Coleoptera	
	*Harmonia axyridis*Pallas.	Coccinellidae: Coleoptera	(95)
	*Hyperaspis bigeminata*Randall.	Coccinellidae: Coleoptera	
	Cybocephalus sppErichson.	Cybocephalidae: Coleoptera	
	Chrysopid	Chrysopidae: Neuroptera	
	Ceraeochrysa spp Adams.	Chrysopidae: Neuroptera	
	Psyllobora parvinotata Casey.	Coccinellidae: Coleoptera	
	Mallada spp Navas.	Chrysopidae: Neuroptera	(61)
	Dichochrsa astur Banks	Chrysopidae: Neuroptera	(7)
	Stethorus spp Weise.	Coccinellidae: Coleoptera	
	Menochilus sexmaculatus F.	Coccinellidae: Coleoptera	
	Chilocorus nigrita Fabricius.	Coccinellidae: Coleoptera	
	Scymnus nubilis Mulsant.	Coccinellidae: Coleoptera	
	Chrysoperla zastrowi sillemi Esben – Petersen.	Chrysopidae: Neuroptera	
	Coccinella transversalis F.	Coccinellidae: Coleoptera	(8)
	Mallada desjardinsi Navas.	Chrysopidae: Neuroptera	
	Propylea dissecta Mulsant.	Coccinellidae: Coleoptera	
	Scymnus saciformis Motschulsky.	Coccinellidae: Coleoptera	
	Pseudomallda astur Banks.	Chrysopidae: Neuroptera	(14)
	Cybocephalus indicus Tian & Ramani.	Nitidulidae: Coleoptera	
	Mallada astur Banks.	Chrysopidae: Neuroptera	
	Mallada boninensis Okamoto.	Chrysopidae: Neuroptera	(15)
	Cheilomenes sexmaculata Fab.	Coccinellidae: Coleoptera	
	Curinus coeruleus Mulsant.	Coccinellidae: Coleoptera,	
	Oecophylla smaragdina Fab.	Formicidae: Hymenoptera	(45)
	Coccinella transversalis Fabricius.	Coccinellidae: Coleoptera	
	2) Bondar's nesting whitefly, *Paraleyrodes bondari*		
	Chrysopids	Chrysopidae: Neuroptera	(25)
	Coccinellids	Coccinellidae: Coleoptera	
	3) Nesting whitefly, *Paraleyrodes minei*		
	*Clitostetus arcuatus*Rossi.	Coccinellidae: Coleoptera	(43)
	*Serangium parcesetosum*Sicard.	Coccinellidae: Coleoptera	
	Coccinellids	Coccinellidae: Coleoptera	
	*Dichochrysa*spp	Chrysopidae: Neuroptera	(13)
	Psocids	Psocidae: Psocoptera	
	4) Palm infesting whitefly, *Aleurotrachelus atratus*		
	Dichochrysa astur Banks.	Neuroptera: Chrysopidae	
	Jauravia pallidula Motschulsky.	Coccinellidae: Coleoptera	(14)
	Chilocorus nigrita Fabricius.	Coccinellidae: Coleoptera	
	Cybocephalus sppErichson.	Cybocephalidae: Coleoptera	
	Parasitoids of invasive whiteflies		
	Rugose spiralling whitefly, *Aleurodicus rugioperculatus*		
	Encarsia guadeloupae Viggiani.	Aphelinidae: Hymenoptera	
	*Eloria noyesi*Schaus.	Erebidae: Lepidoptera	(95)
	Aleuroctonus vittatus Dozier.	Eulophidae: Hymenoptera	
	Encarsia dispersa Polaszek	Aphelinidae: Hymenoptera	(97)
	Encarsia fernandae Sanchez & Myartseva.	Aphelinidae: Hymenoptera	(108)
	2) Nesting whitefly, *Paraleyrodes minei*		
	Encarsia dominicana Evans.	Aphelinidae: Hymenoptera	
	Encarsiaparvella Silvestri.	Aphelinidae: Hymenoptera	(43)
	Encarsiavariegata Howard.	Aphelinidae: Hymenoptera	
	Encarsia spp Foerster.	Aphelinidae: Hymenoptera	(18)
	3) Palm infesting whitefly, *Aleurotrachelus atratus*		
	Eretmocerus cocois Delvare sp	Aphelinidae: Hymenoptera	(109)
	Encarsia basicinta	Aphelinidae: Hymenoptera	
	Encarsia spp Foerster.	Aphelinidae: Hymenoptera	(110)
	Signiphora spp Howard.	Signiphoridae: Hymenoptera	
	*Encarsia cubensis*Gahan.	Aphelinidae: Hymenoptera	(111)

Table 4. Annual scientific production of invasive whiteflies

Year	Articles
1996	1
1997	0
1998	1
1999	0
2000	0
2001	0
2002	0
2003	0
2004	1
2005	0
2006	0
2007	0
2008	1
2009	0
2010	1
2011	2
2012	3
2013	2
2014	3
2015	3
2016	8
2017	7
2018	11
2019	7
2020	15
2021	23
2022	30
2023	31
2024	19

Table 5. Average citations per year of invasive whiteflies

Year	MeanTC per Art	N	MeanTC per year	Citable years
1996	21	1	0.72	29
1998	9	1	0.33	27
2004	3	1	0.14	21
2008	2	1	0.12	17
2010	31	1	2.07	15
2011	5.5	2	0.39	14
2012	9	3	0.69	13
2013	12.5	2	1.04	12
2014	2	3	0.18	11
2015	7	3	0.7	10
2016	10.38	8	1.15	9
2017	13.14	7	1.64	8
2018	4.18	11	0.6	7
2019	9.14	7	1.52	6
2020	3.33	15	0.67	5
2021	4.04	23	1.01	4
2022	2.1	30	0.7	3
2023	0.65	31	0.32	2
2024	0.11	19	0.11	1

Table 6. Most relevant sources of are invasive whiteflies

Sources (Journal name)	Articles
Indian Journal of Entomology	12
Phytoparasitica	12
Research Square	9
International Journal of Tropical Insect Science	7
Journal of Biological Control	7
Journal of Plantation Crops	5
CABI Compendium	4
Distribution Maps of Plant Pests	4
EDIS	4
Entomon	4
Florida Entomologist	4
IOP Conference Series Earth and Environmental Science	4
Journal of Economic Entomology	3
Pest Management in Horticultural Ecosystems	3
Southwestern Entomologist	3

Table 7. Most relevant authors of invasive whiteflies

Authors	Articles	Articles Fractionalized
Selvaraj K	15	3.77
Josephrajkumar A	11	1.77
Hedge V	8	1.38
Babu M	7	1.13
Sundararaj R	7	1.97
Elango K	6	2.25
Gupta A	6	2.04
Mannion C	6	1.75
Sumalatha BV	6	1.58
Mohan C	5	0.72

Table 9. Country-wise scientific production of invasive whiteflies

Region	Freq
India	49
USA	14
UK	8
Bangladesh	6
France	2
Greece	2
Indonesia	2
Italy	2
Tanzania	2
China	1
Costa Rica	1
Japan	1
Kenya	1
EU	1
Senegal	1
Spain	1
Sri Lanka	1
Turkey	1
Uganda	1

Table 8. Globally cited documents of invasive whiteflies

Paper	DOI	Total citations	TC per year	Normalized TC
Sundararaj R, 2017, Phytoparasitica	10.1007/S12600-017-0567-0	54	6.75	4.11
Francis AW, 2016, Florida Entomologist	10.1653/024.099.0134	38	4.22	3.66
Boroweic N, 2010, Journal of Applied Entomology	10.1111/J.1439-0418.2009. 01450.X	31	2.07	1.00
Shanas S, 2016, Entomon	10.33307/ENTOMON.V41I4.227	28	3.11	2.70
Maruthadurai R, 2022, Pest Management Science	10.1002/PS.7199	22	7.33	10.48
Martin J, 1996, Journal of Natural History	10.1080/00222939600771081	21	0.72	1.00
Selvaraj K, 2017, Journal of Biological Control	10.18311/JBC/2017/16015	20	2.50	1.52
Kumar V, 2018, Journal of Economic Entomology	10.1093/JEE/TOY056	19	2.71	4.54
Sundararaj R, 2021, Phytoparasitica	10.1007/S12600-021-00919-7	18	4.50	4.45
Josephrajkumar A, 2019, Phytoparasitica	10.1007/S12600-019-00741-2	17	2.83	1.86

Additionally, intercropping with non-host plants can make the environment less conducive to whitefly infestations, reducing their spread (73). Physical and mechanical methods are also effective, especially in the early stages of infestation. For instance, placing yellow sticky traps around coconut plantations helps monitor and capture adult whiteflies, which are drawn to the yellow colour and become trapped (74).

Washing infested leaves with a strong jet of water is another practical method to knock down whiteflies, particularly when combined with other control strategies (61). Fungal pathogens like *Beauveria bassiana* and *Isaria fumosorosea* effectively control whiteflies by infecting and killing them (75). While the development of whitefly-resistant coconut varieties is ongoing, breeding programs aim to provide a long-term solution for managing these pests (76). Horticultural oils and neem-based products are effective against whitefly nymphs, with neem oil offering both repellent and insecticidal properties (61). Insect growth regulators (IGRs), like buprofezin, target immature whitefly stages, reducing future populations while sparing beneficial organisms (77). Systemic insecticides, such as imidacloprid and thiamethoxam, can control whiteflies, but should be used sparingly to prevent resistance (78). Sooty mould, a secondary issue caused by honeydew secretions, can be managed by controlling whiteflies and cleaning affected leaves with water or a mild soap solution to restore photosynthesis (69).

The parasitoid *Encarsia noyesi* effectively reduced whitefly survival, with heavy treatment resulting in less than 10 % survival to adulthood (79). Studies on the natural parasitization of *Encarsia guadeloupae* on rugose spiraling whitefly revealed the highest parasitization rate in Kanyakumari district (33.86 %) during the study period. The percentage of nymphal parasitization by *E. guadeloupae* was observed in the following order: Kanyakumari (33.86 %) > Tirunelveli (17.66 %) > Thoothukudi (12.45 %) > Tenkasi (11.20 %). The highest adult emergence rate of *E. guadeloupae* (21.41 %) was recorded in coconut gardens of Kanyakumari, while the lowest was in Tenkasi district (5.37 %) (80). The successful introduction of *E. guadeloupae* as a biological control agent demonstrated its effectiveness, mainly when supported by conservation practices (81).

The predatory potential of the chrysopid predator *Pseudomallda astur* against rugose spiralling whitefly was investigated, showing that as larval instars progressed, whitefly egg consumption increased significantly, with first instar larvae consuming 94.75 % of 80 eggs (83). Releasing *Apertochrysa astur* at 600 eggs per palm effectively minimized RSW populations (84). Further exploration of *A. astur* predatory efficiency revealed that third-instar grubs could consume up to 333 second-instar nymphs of BNW, highlighting their potential as a biological control agent for managing whitefly populations (85).

The third instar grub of *Cryptolaemus montrouzieri* was shown to consume the highest number of whiteflies in the shortest time (11). During their development, grubs of *Chrysoperla zastrowi sillemi* and *Mallada boninensis* consumed an average of 653.6 and 929.8 whiteflies, respectively (46). The predatory capacity of *Menochilus sexmaculatus* was evaluated at different temperatures, with the highest consumption observed at 27 ± 1°C; adult females consumed more prey than males, highlighting the species' adaptability and effectiveness in managing whitefly populations (86). *Nephaspis oculata* was identified as an effective predatory lady beetle for controlling rugose spiraling whitefly, consuming an average of 245.7 eggs before pupation (87).

A 50 % mortality rate of whiteflies was achieved using the fungal biocontrol agent *Lecanicillium lecanii*. Botanical controls were also evaluated, with azadirachtin 5 % showing the highest mortality rates 66.67 % for nymphs and 70 % for adults (15). The fungus *Simplicillium lanosoniveum* was characterized and found to be highly virulent against whitefly eggs and nymphs, marking the first recorded instance of this fungus infecting RSW and highlighting its potential as a mycoinsecticide (88).

Two applications of the entomopathogenic fungus *Isaria fumosorosea* significantly reduced whitefly populations in coconut gardens, achieving mortality rates of up to 97.03 %. Control efficacy was further enhanced when combined with the parasitoid *Encarsia guadeloupae* (89). *I. fumosorosea* was highlighted as highly effective against whitefly populations, with reductions reaching up to 100 %. Additionally, combining the insect growth regulator buprofezin with *I. fumosorosea* demonstrated notable effectiveness in Florida (90). Evaluation of a specific *I. fumosorosea* strain revealed varying mortality rates across different whitefly life stages (91). Among several bio-pesticides tested, *I. fumosorosea* was the most effective (92) and its combination with chemical insecticides significantly reduced whitefly infestations (93).

A combination of *Isaria fumosorosea* (ICAR-NBAIR Pfu-5) and profenophos significantly reduced rugose spiralling whitefly (RSW) infestations, achieving reductions of 82.97 % in infestation levels and 79.68 % in mean live colonies. A similar combination with buprofezin also proved effective (92). Various insecticidal treatments were evaluated, with Nitro 505 EC and Bioclean yielding the best results, the latter being highlighted for its eco-friendliness (94). An integrated pest management (IPM) approach tested in Maharashtra demonstrated that neem oil, water sprays and yellow sticky traps effectively lowered RSW populations (59). Acetamiprid was identified as the most effective treatment, with a rotation strategy involving bio-pesticides like D-Lemonine recommended for sustainable management (57). Additionally, soap nut treatments effectively controlled RSW, supporting eco-friendly pest management alternatives (59).

The toxicity of methanol extracts from Sweet Flag (*Acorus calamus*) against Bondar's nesting whitefly (*Paraleyrodes bondari*) was evaluated, revealing that mortality rates increased with higher concentrations, reaching 100 % at 5 %. The LC_{50} value was determined to be 0.470 % at a 0.5 % concentration (95). Monitoring whitefly flight patterns was highlighted as crucial for effective pest management. Rugose spiralling whitefly (RSW) activity peaked in the morning, particularly in the Southeast, while Bondar's nesting whitefly (BNW) exhibited a similar preference for that time and location, with minimal evening activity (96).

Conclusion

Invasive whiteflies threaten coconut ecosystems, driven by their wide geographic distribution, adaptability to various host plants and the substantial damage they inflict on coconut palms. By feeding on phloem sap, these pests disrupt the plant's nutrient transport system, resulting in symptoms such as yellowing leaves, premature leaf drop and overall reduced vigour. The rapid spread of whiteflies is facilitated by environmental conditions favouring their proliferation, including warm temperatures and high humidity. Their ability to infest various plant species complicates management efforts, as these pests can easily migrate between hosts. Additionally, the excretion of honeydew by whiteflies leads to the growth of sooty mould, further obstructing photosynthesis and exacerbating the decline in plant health and productivity. Accurate taxonomic identification of the various whitefly species is vital for developing targeted control measures, as different species may exhibit distinct behaviours and vulnerabilities. Identifying invasive whitefly species remains a complex challenge due to their morphological similarities. Therefore, systematic morphological analysis using taxonomic keys, complemented by molecular markers, is crucial for effective pest identification. The increasing prevalence of these invasive whitefly species underscores the urgent need for comprehensive and proactive pest management strategies to safeguard coconut plantations. Combining biological, cultural and chemical control methods within an IPM framework can mitigate whitefly infestations and protect the productivity of coconut palms. Continued research and collaboration among growers, agricultural experts and researchers will be pivotal in developing innovative solutions to combat the challenges of invasive whiteflies, ultimately fostering resilience within coconut ecosystems.

Acknowledgements

The authors thank Tamil Nadu Agricultural University for supporting this experiment. They also sincerely thank the chairperson and advisory committee for their valuable guidance and contributions during the preparation of this review.

Authors' contributions

SS, GP and SV conducted literature searches and data extraction and analyzed and interpreted the compiled information. NB and JS conceptualized the review topic, provided guidance on the review process and approved the final manuscript. GM and AA contributed to manuscript editing, summarization and revision.

Compliance with ethical standards

Conflict of interest: Authors do not have any conflict of interest to declare.

Ethical issues: None

References

1. Zhang Y, Kan J, Liu X, Song F, Zhu K, Li N, Zhang Y. Chemical components, nutritional value, volatile organic compounds and biological activities *in vitro* of coconut (*Cocos nucifera* L.) water with different maturities. Foods. 2024;13(6):863. https://doi.org/10.3390/foods13060863
2. International Coconut Community. Statistical Year Book. International Coconut Community; 2021
3. Coconut Development Board (2022-23). Area, production and productivity of coconut in India. CDB; 2024 [cited 2025 Feb 20]. Available from: https://coconutboard.gov.in/Statistics.aspx
4. Abhishek TS, Dwivedi SA. Review on integrated pest management of coconut crop. Int J Entomol Res. 2021;6:115–20.
5. Dubey AK, Sundararaj R. A new combination and first record of the genus *Aleurothrixus* Quaintance and Baker (Hemiptera: Aleyrodidae) from India. Biosystematica. 2015;9(1-2):23–28.
6. Shanas S, Joseph J, Tom J, Anju KG. First report of the invasive rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) from the old world. Entomon. 2016;41 (4):365–68. https://doi.org/10.33307/entomon.v41i4.227
7. Selvaraj K, Gupta A, Venkatesan T, Jalali SK, Sundararaj R, Ballal CR. First record of invasive rugose spiraling whitefly *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) along with parasitoids in Karnataka. J Biol Control. 2017;31(2):74–78. https://doi.org/ 10.18311/jbc/2017/16015
8. Srinivasan T, Saravanan PA, Josephrajkumar A, Sridharan S, David PM. Invasion of the rugose spiralling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera Aleyrodidae) in Pollachi tract of Tamil Nadu, India. Madras Agric J. 2016;103(10-12):349–53. https://doi.org/10.29321/MAJ.10.001047
9. Josephrajkumar A, Mohan CH, Krishnakumar V. Parasitism induced bio-suppression of coconut whitefly in Kerala. Kerala Karshakan. 2016;4(7):26–27.
10. Mohan CCM, Josephrajkumar A, Hegde VVH, Krishnakumar V, Renjith PB, Anjali AS, Chowdappa A. Gradient out break and bio-suppression of spiralling whitefly in coconut gardens in South India. Indian Coconut J. 2016;59 (8):9–12.
11. Elango K, Nelson S, Sridharan S, Paranidharan V, Balakrishnan S. Biology, distribution and host range of new invasive pest of India coconut rugose spiralling whitefly *Aleurodicus rugioperculatus* Martin in Tamil Nadu and the status of its natural enemies. Int J Agric Sci. 2019;11(9):8423–26.
12. Chandrika M, Josephrajkumar A, Merin B, Arya K, Krishnakumar V. Occurrence of invasive Bondar's nesting whitefly on coconut in Kerala. Indian Coconut J. 2019;61(9):17–18.
13. Sujithra M, Prathibha VH, Hegde V, Poorani J. Occurrence of nesting whitefly *Paraleyrodes minei* iaccarino (Hemiptera: aleyrodidae) in India. Indian J Entomol. 2019;81(3):507–10. https://doi.org/10.5958/0974-8172.2019.00109.3
14. Selvaraj K, Sundararaj R, Sumalatha BV. Invasion of the palm infesting whitefly, *Aleurotrachelus atratus* Hempel (Hemiptera: Aleyrodidae) in the oriental region. Phytoparasitica. 2019;47:327–32. https://doi.org/10.1007/s12600-019-00742-1
15. Elango K, Nelson SJ. Influence of intercrops in coconut on *Encarsia guadeloupae* Viggiani parasitization of rugose spiralling whitefly *Aleurodicus rugioperculatus* Martin. Ann Plant Prot Sci. 2020;28(1):1–4. https://doi.org/10.5958/0974-0163.2020.00001.4
16. Vidya CV, Sundararaj R, Dubey AK, Bhaskar H, Chellappan M, Henna MK. Invasion and establishment of Bondar's nesting whitefly, *Paraleyrodes bondari* Peracchi (Hemiptera: Aleyrodidae) in Indian mainland and Andaman and Nicobar Islands. Entomon. 2019;44(2):149–54. https://doi.org/10.33307/entomon.v44i2.443
17. Dhaliwal GS, Arora R. Role of phytochemicals in integrated pest management. Int Phytochem Biopest. 2000:92–109. https://

doi.org/10.1201/9780203304686-18

18. Evans GA. The whiteflies (Hemiptera: Aleyrodidae) of the world and their host plants and natural enemies. USDA; 2008.
19. Stocks IC. Bondar's Nnesting whitefly, *Paraleyrodes bondari*, a whitefly (Hemiptera: Aleyrodidae) new to Florida attacking ficus and other hosts. Pest Alert; 2012.
20. Rao NC, Roshan DR, Rao GK, Ramanandam G. A review on rugose spiralling whitefly, *Aleurodicus rugioperculatus* martin (Hemiptera: Aleyrodidae) in India. J Pharmacogn Phytochem. 2018;7(5):948–53.
21. Chakravarthy AK, Kumar KP, Sridhar V, Prasannakumar NR, Nitin KS, Nagaraju DK, et al. Incidence, hosts and potential areas for invasion by rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) in India. Pest Manag Hort Ecosys. 2017;23(1):41–49.
22. Mondal P, Ganguly M, Bandyopadhyay P, Karmakar K, Kar A, Ghosh DK. Status of rugose spiraling whitefly *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) in West Bengal with notes on host plants, natural enemies and management. J Pharmacogn Phytochem. 2020;9(1):2023–27.
23. Jethva DM, Wadaskar PS, Kachot AV. First report of rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) on coconut in Gujarat, India. J Entomol Zool Stud. 2020;8:722–25.
24. Omongo CA, Namuddu A, Okao-Okuja G, Alicai T, van Brunschot S, Ouvrard D, Colvin J. Occurrence of Bondar's nesting whitefly, *Paraleyrodes bondari* (Hemiptera: Aleyrodidae), on cassava in Uganda. Rev Bras Entomol. 2018;62:2579. https://doi.org/10.1016/j.rbe.2018.10.001
25. Josephrajkumar A, Mohan C, Babu M, Krishna A, Krishnakumar V, Hegde V, Chowdappa P. First record of the invasive Bondar's nesting whitefly, *Paraleyrodes bondari* Peracchi on coconut from India. Phytoparasitica. 2019;47(3):333–39. https://doi.org/10.1007/s12600-019-00741-2
26. Banumathi K, Murugan M, Jeyarani S, Mohankumar S, Balasubramani V, Sowmiya C. Prevalence of invasive Aleyrodidae harbouring horticultural host plants in different ecosystems of Tamil Nadu. J Entomol Zool Stud. 2020;8(6):886–90. https://doi.org/10.22271/j.ento.2020.v8.i6l.7955
27. Martin JH. Whiteflies of Belize (Hemiptera: Aleyrodidae). Part 1 introduction and account of the subfamily Aleurodicinae Quaintance and baker. Zootaxa. 2004;681(1):1–19. https://doi.org/10.11646/zootaxa.681.1.1
28. Liu B, Qin WQ, Yan W. Potential geographical distribution of *Paraleyrodes minei* (Hemiptera; Aleyrodidae) in China based on Maxent model. J Environ Entomol. 2019;41(6):1276–86. https://doi.org/10.3969/j.issn.1674-0858.2019.06.17
29. Kalaitzaki AP, Tsagkarakis AE, Ilias A. First record of the nesting whitefly, *Paraleyrodes minei*, in Greece. Entomol Hell. 2016;25(1):16–21. https://doi.org/10.12681/eh.11547
30. Dubey AK. *Paraleyrodes minei* Iaccarino (Hemiptera: Aleyrodidae)-a new invasive pest threat to Andaman and Nicobar Islands, India. Phytopara. 2019;47(5):659–62. https://doi.org/10.1007/s12600-019-00760-z
31. Howard FW, Giblin-Davis R, Moore D, Abad R. Insects on palms. Cabi; 2001. https://doi.org/10.1079/9780851993263.0000
32. Beaudoin-Ollivier L, Streito JC, Ollivier J, Delvare G, Julia JF, Ryckewaert P, Ali M. *Aleurotrachelus atratus* hempel (Hemiptera: Aleyrodidae) and its emergence as a pest of coconut (*Cocos nucifera* L.) in the Comoro Islands. In: 2nd European Whitefly Symposium [EWSII], [5-9 October 2004], Cavtat, Croatia; 2004. pp. 17–18.
33. Borowiec N, Quilici S, Martin J, Issimaila MA, Chadhouliati AC, Youssoufa MA, et al. Increasing distribution and damage to palms by the neotropical whitefly, *Aleurotrachelus atratus* (Hemiptera: Aleyrodidae). J Appl Entomol. 2010;134(6):498–510. https://doi.org/10.1111/j.1439-0418.2009.01450.x
34. Malumphy C, Reseder K. Palm-infesting whitefly *Aleurotrachelus atratus* (Hempel) (Hemiptera: Aleyrodidae) established in England at a botanical garden. Entomol Mon Mag. 2011;147(1760-62):23–31.
35. Selvaraj K, Sumalatha BV, Sundararaj R. New distributional record of invasive neotropical coconut whitefly *Aleurotrachelus atratus* (Hemiptera: Aleyrodidae) in Tamil Nadu, India. Insect Environ. 2021;24(2):230–35.
36. Sajan JV, Prathibha PS, Diwakar Y, Josephrajkumar A. New distribution record of palm whitefly, *Aleurotrachelus atratus* Hempel in Kerala, India. Indian Coconut J. 2022;65(1):10–12.
37. Quaintance AL, Baker AC. Classification of the Aleyrodidae part II. Technical series, Unites States Department of Agriculture Bureau Entomol. 1914;27:95–109. https://doi.org/10.5962/bhl.title.123077
38. Cockerell TD. The classification of the Aleyrodidae. Proceed Acad Nat Sci Philadel. 1902:279–83.
39. Martin JH, Mifsud D, Rapisarda C. The whiteflies (Hemiptera: Aleyrodidae) of Europe and the Mediterranean basin. Bull Entomol Res. 2000;90(5):407–48. https://doi.org/10.1017/S0007485300000547
40. Selvaraj K, Sumalatha BV, Sundararaj R, Venkatesan T, Shylesha AN, Sushil SN. Guide on diagnosis of invasive whiteflies and their natural enemies. Technical Bulletin 2021;1.
41. Sadhana V, Senguttuvan K, Murugan M. Taxonomy of whiteflies' natural enemies in Tamil Nadu cotton ecosystem. Madras Agricul J. 2022;109(7-9):90–94. https://doi.org/10.29321/MAJ.10.000711
42. Stocks IC, Hodges G. The rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin, a new exotic whitefly in South Florida (Hemiptera: Aleyrodidae). Florida: Department of Agriculture and Consumer Services; 2012.
43. Iaccarino FM, Jesu R, Giacometti R. *Paraleyrodes minei* Iaccarino 1990 (Homoptera: Aleyrodidae), new specie for Italy, on *Citrus aurantium* L. J Entomol Acarol Res. 2011;43(1):1–6. https://doi.org/10.4081/jear.2011.1
44. Yasodha P, Fousiya A, Elakkiya K, Justin CGL, Masilamani P. Comparative studies on exotic neotropical whiteflies of coconut. J Entomol Zool Stud. 2020;8(3):627–29.
45. Alagar M, Rajamanikam K, Chinnadurai S, Yasmin A, Maheswarappa HP. Surveillance, assessment of infestation, biology, host range of an invasive rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin and status of its natural enemies in Tamil Nadu. J Entomol Zool Stud. 2020;8(3):2041–47.
46. Elango K, Nelson SJ, Sridharan S, Paranidharan V, Balakrishnan S. Biology and predatory potential of chrysopids on invasive coconut rugose spiralling whitefly *Aleurodicus rugioperculatus* Martin. Indian J Exp Biol. 2021;59(11):814–18.
47. Suriya S, Preetha G, Balakrishnan N, Sheela J. Seasonal incidence, population dynamics and morphometric traits of exotic coconut whiteflies in southern Tamil Nadu. J Hortic Sci. 2023;18(1):216–22. https://doi.org/10.24154/jhs.v18i1.2167
48. Anand G, Kathirvelu C, Ayyasamy R. Comparison of biology and reproductive potential of two invasive coconut whiteflies, *Aleurodicus rugioperculatus* Martin and *Paraleyrodes bondari* Peracchi through an age-stage, two-sex life table approach. Uttar Pradesh J Zool. 2023;44(23):52–60. https://doi.org/ 10.56557/UPJOZ/2023/v44i233765
49. Sadhana V, Senguttuvan K, Murugan M, Boopathi MN, Sathiah N. First record of Bondar's nesting whitefly, *Paraleyrodes bondari* Peracchi (Hemiptera: Aleyrodidae), occurrence and infestation in the cotton ecosystem of Tamil Nadu, India. Pharm Innov. 2021;10(10S):1278–84.
50. Pradhan SK, Shylesha AN, Selvaraj K, Sumalatha VB. Comparative biology of invasive rugose spiralling whitefly *Aleurodicus rugioperculatus* Martin on three host plants. Indian J Entomol. 2020;82(3):498–503. https://doi.org/10.5958/0974-8172.2020.00125.X

51. Sankarganesh E, Roy K, Ali MN. Distribution and identification of invasive whiteflies on coconut palms in eastern India and its natural enemies: A biosecurity perspective. In: IOP Conference Series: Earth Environ Sci. 2023;1179(1):012005. https://doi.org/10.1088/1755-1315/1179/1/012005

52. Raghuteja PV, Rao NB, Padma E, Neeraja B, Kireeti A, Rao VG, et al. Host dynamics and molecular characterization of neo tropical invasive Bondar's nesting whitefly (BNW), *Paraleyrodes bondari* Peracchi (Hemiptera: Aleyrodidae) in Andhra Pradesh. Pest Manag Hort Ecosys. 2022;28(2):103–09. http://dx.doi.org/10.5958/0974-4541.2022.00046.7

53. Sundararaj R, Krishnan S, Sumalatha BV. Invasion and expansion of exotic whiteflies (Hemiptera: Aleyrodidae) in India and their economic importance. Phytopara. 2021;49(5):851–63. https://doi.org/10.1007/s12600-021-00919-7

54. Chakraborty D, Sahoo SK. First report of three invasive whitefly species (Aleyrodidae: Hemiptera) from West Bengal, India. Int J Environ Clim Chang. 2023;13(8):939–44. https://doi.org/10.9734/ijecc/2023/v13i82031

55. Heersmink R, van den Hoven J, van Eck NJ, van den Berg J. Bibliometric mapping of computer and information ethics. Ethics Inf Technol. 2011;13:241–49. https://doi.org/10.1007/s10676-011-9273-7

56. Tang M, Liao H, Wan Z, Herrera-Viedma E, Rosen M. Ten years of sustainability (2009 to 2018): A bibliometric overview. Sustain. 2018;10:1655. https://doi.org/10.3390/su10051655

57. Dutta NK, Sarker D, Begum K, Sarkar MA, Islam MI, Rahman MM. First record of the invasive rugose spiraling whitefly, *Aleurodicus rugioperculatus* martin (hemiptera: Aleyrodidae) in Bangladesh with its host range and status as coconut pest. Bangladesh J Entomol. 2019;29(2):73–83.

58. Das G, Uddin MM, Roy BK, Ahemed MS. Identification of rugose spiralling whitefly infesting host plants in Bangladesh and assessment of its damage severity. Am Entomol. 2023;7(1):1–8. https://doi.org/10.11648/j.aje.20230701.11

59. Rahman MM, Dutta NK, Sarkar MA, Nuruzzaman M, Islam MR. Surveillance and biorational management of rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) infesting coconut to reduce the invasion threat in Bangladesh. 2024 (preprint). https://doi.org/10.21203/rs.3.rs-3859422/v1

60. Puvvala VR, Nagulapati BV, Ede P, Alwala K, Nathala E, Kavuru U. Induced nut yield reduction in Godavari Ganga hybrid coconut palms: Invasive rugose spiraling whitefly (*Aleurodicus Rugioperculatus* Martin) infestation. Appl Fruit Sci. 2024;66(2):755–62. https://doi.org/10.1007/s10341-024-01054-3

61. Wankhede SM, Shinde VV, Ghavale SL. Status of rugose spiralling whitefly (*Aleurodicus rugioperculatus* Martin) in Konkan region of Maharashtra. Pest Manag Hort Ecosys. 2021;27(2):190–95.

62. Cugala D, Alfredo J, Mataruca M, Chiconela T, Muthammbe A, Martins C, Majacunene A. Assessment of severity infestation of the coconut whitefly, *Aleurotrachelus atratus* and its associated impact on coconut productivity in Inhambane province, Mozambique. In: 11th African Crop Science Proceedings, Sowing innovations for sustainable food and nutrition security in Africa. Entebbe, Uganda; 2013. p. 273–77.

63. Sundararaj R, Selvaraj K. Invasion of rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae): a potential threat to coconut in India. Phytoparasitica. 2017;45:71–74. https://doi.org/10.1007/s12600-017-0567-0

64. Patel SS, Sisodiya DB, Parmar RG. "Rugose spiralling whitefly (RSW): An invasive pest in India. J Entomol Zool Stud. 2020;8(6):1865–67.

65. Sushmitha S, Sujatha A, Emmanuel N, Krishna KU, Suneetha DR. Incidence, intensity and host dynamics of rugose spiralling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) in Andhra Pradesh. Pest Manag Hort Ecosys 2020;26(1):109–20. https://doi.org/10.5958/0974-4541.2020.00018.1

66. Pradhan SK, Shylesha AN, Selvaraj K, Sumalatha BV. Distribution, host range and status of invasive rugose spiralling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) in Karnataka. Agric Res. 2021;11:1–7. https://doi.org/10.1007/s40003-021-00593-5

67. Chavan SS, Narangalkar A, Sapkal SD. Population dynamics of rugose spiralling whitefly, *Aleurodicus rugioperculatus* (Martin) on coconut. Pharm Innov J. 2022;SP-11(3):864–67.

68. Chandrasekaran G, Subramanian J, Marimuthu M, Subbarayalu M, Shanmugam H. Invasive whitefly (Hemiptera: Aleyrodidae) complex and diversity in coconut landscapes in Tamil Nadu1. J Entomol Sci. 2023;58(4):388–99. https://doi.org/10.18474/JES23-17

69. Khan MMH. Abundance and control of rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin, infesting coconut in seven coastal districts of Bangladesh: Abundance and control of rugose spiraling whitefly. J Asiat Soc Bangladesh Sci. 2023;49(2):165–77. https://doi.org/10.3329/jasbs.v49i2.70766

70. Gerling D, Alomar Ò, Arnò J. Biological control of *Bemisia tabaci* using predators and parasitoids. Crop Prot. 2001;20(9):779–99. https://doi.org/10.1016/S0261-2194(01)00111-9

71. Ramani N, Josephrajkumar A, Sundararaj R. Management of rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) on coconut. J Plant Crops. 2012;40(1):23–28.

72. Sujatha A, Sundararaj R, Mani, M. Integrated pest management of coconut whiteflies. J Hortic Sci. 2017;12(2):115–20.

73. Mitchell PL, Sengonca C. Whitefly pests: Cotton and other crops. CABI Publishing; 1997.

74. Jones DR. Plant viruses transmitted by whiteflies. Eur J Plant Pathol. 2003;109:195–219. https://doi.org/10.1023/A:1022846630513

75. Hoddle MS, Van Driesche RG, Sanderson JP, Minkenberg OP. Biological control of *Bemisia argentifolii* (Hemiptera: Aleyrodidae) on poinsettia with inundative releases of *Eretmocerus eremicus* (Hymenoptera: Aphelinidae): do release rates affect parasitism?. Bull Entomol Res.1998;88(1):47–58. https://doi.org/10.1017/S0007485300041547

76. Faria M, Wraight SP. Biological control of *Bemisia tabaci* with fungi. Crop Prot. 2001;20(9):767–78. https://doi.org/10.1016/S0261-2194(01)00110-7

77. Perring TM, Cooper AD, Rodriguez RJ, Farrar CA, Bellows Jr TS. Identification of a whitefly species by genomic and behavioral studies. Sci. 1993;259(5091):74–77. https://doi.org/ 10.1126/science.8418497

78. Naranjo SE, Ellsworth PC. The contribution of conservation biological control to integrated control of *Bemisia tabaci* in cotton. Biol Control. 2009;51(3):458–70. https://doi.org/10.1016/j.biocontrol.2009.08.006

79. Horowitz AR, Ishaaya I. Insect pest management: Field and protected crops. Springer; 2004. https://doi.org/10.1007/978-3-662-07913-3

80. Boughton AJ, Mendez MA, Francis AW, Smith TR, Osborne LS, Mannion CM. Host stage suitability and impact of *Encarsia noyesi* (Hymenoptera: Aphelinidae) on the invasive rugose spiraling whitefly, *Aleurodicus rugioperculatus* (Hemiptera: Aleyrodidae), in Florida. Biol Control. 2015;88:61–67. https://doi.org/10.1016/j.biocontrol.2015.04.016

81. Suriya S, Preetha G, Balakrishnan N, Sheela, J. Natural parasitization of aphelinid parasitoid, *Encarsia guadeloupae* Viggiani on coconut rugose spiralling whitefly, *Aleurodicus rugioperculatus* Martin. Pharm Innov J. 2021;SP-10(11):388–90.

82. Rao NC, Anoosha V, Neeraja B, Rao VG, Kireeti A, Rao GK,

Srinivasulu B. Conservational biological control approach of rugose spiraling whitefly. Indian Coconut J. 2023;65(9):22–24.

83. Rao NC, Raman B, Bhagavan BV. Functional response and density dependent feeding interaction of *Pseudomallada astur* Banks (Neuroptera: Chrysopidae) against rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae). Pest Manag Hort Ecosyst. 2020;26(2):229–34.

84. Raghuteja PV, Rao NC, Padma E, Sekhar V. Evaluation of release rates of predator, *Apertochrysa astur* (Banks) (Neuroptera: Chrysopidae) against rugose spiraling whitefly (RSW), *Aleurodicus rugioperculatus* Martin. J Biol Control. 2023;37(1):26–31. https://doi.org/10.18311/jbc/2023/33539

85. Remoniya X, Jeyarajan NS, Jeyarani S, Mohankumar S, Sivakumar U, Chitra N. Biology of green lacewing, *Apertochrysa astur* (Banks) (Neuroptera: Chrysopidae) and its predatory potential against invasive rugose spiralling whitefly, *Aleurodicus rugioperculatus* Martin and Bondar's nesting whitefly, *Paraleyrodes bondari* Peracchi (Hemiptera: Aleyrodidae) of coconut. Int J Trop Insect Sci. 2024;44(1):9–104. https://doi.org/10.1007/s42690-023-01112-5

86. Anjum H, Ali M. Predatory potential of coccinellid beetle, *Menochilus sexmaculatus* Fabricius (Coleoptera) at different temperatures, against rugose spiralling whitefly *Aleurodicus Rugioperculatus* Martin (Hemiptera). Mun Ent Zool. 2021;6:1070–74.

87. Taravati S, Mannion C, Osborne LS. Management of rugose spiraling whitefly (*Aleurodicus rugioperculatus*) in the South Florida landscape. In: Proceedings of the Florida State Horticultural Society, 2013;126:276–78. https://doi.org/10.32473/edis-in1004-2013

88. Sujithra M, Prathibha HV, Rajkumar M, Guru-Pirasanna-Pandi G, Senthil-Nathan S, Hegde V. Entomopathogenic potential of *Simplicillium lanosoniveum* native strain in suppressing invasive whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae), infesting coconut. J Fungi. 2021;7(11):964. https://doi.org/10.3390/jof7110964

89. Visalakshi M, Selvaraj K, Sumalatha BP, Poornesha B. Biological control of invasive pest, rugose spirallying whitefly in coconut and impact on environment. J Entomol Zool Stud. 2021;9(1):1215–18.

90. Kumar V, Francis A, Avery PB, McKenzie CL, Osborne LS. Assessing compatibility of *Isaria fumosorosea* and buprofezin for mitigation of *Aleurodicus rugioperculatus* (Hemiptera: Aleyrodidae): An invasive pest in the Florida landscape. J Econ Entomol. 2018;111 (3):1069–79. https://doi.org/10.1093/jee/toy056

91. Sumalatha BV, Selvaraj K, Poornesha B, Ramanujam B. Pathogenicity of entomopathogenic fungus *Isaria fumosorosea* on rugose spiralling whitefly *Aleurodicus rugioperculatus* and its effect on parasitoid *Encarsia guadeloupae*. Biocontrol Sci Technol. 2020;30(10):1150–61. https://doi.org/10.1080/09583157.2020.1797632

92. Reddy MJ, Chalapathirao NBV, Viji CP, Narashimarao S. Evaluation of biopesticides against rugose spiraling whitefly *Aleurodicus rugioperculatus* Martin on coconut (*Cocos nucifera* L.). Pharm Innov J. 2022;11(8):2108–10.

93. Sandeep A, Selvaraj K, Kalleshwaraswamy CM, Hanumanthaswamy BC, Mallikarjuna HB. Field efficacy of *Isaria fumosorosea* alone and in combination with insecticides against *Aleurodicus rugioperculatus* on coconut. Egypt J Biol Pest Control. 2022;32(1):106. https://doi.org/10.1186/s41938-022-00600-z

94. Khan MMH. Host range of rugose spiraling whitefly, (*Aleurodicus rugioperculatus*) martin and its incidence and damage on coconut plant in coastal region of Bangladesh. SAARC J Agric. 2022:183–97. https://doi.org/10.3329/sja.v20i2.63580

95. Meenakshi G, Emmanuel N, Rammiah DA, Swami DV. Toxic effect of sweet flag *Acorus calamus* extracts against invasive pest Bondar nesting whitefly *Paraleyrodes bondari* Peracchi. (Aleyrodidae, Hemiptera). Pharm Innov J. 2022;11(8S):1328–30.

96. Logeshkumar P, Nalini R, Rajkumar AJ, Chandramani P, Mini ML, Singh RD, Murugan M. Flight activity of invasive rugose spiralling whitefly (*Aleurodicus rugioperculatus* Martin) and bondars nesting whitefly (*Paraleyrodes bondari* Perrachi) in coconut orchards. Res Crops. 2023;24(2):403–06. https://doi.org/10.31830/2348-7542.2023.ROC-946

97. Francis AW, Stocks IC, Smith TR, Boughton AJ, Mannion CM, Osborne LS. Host plants and natural enemies of rugose spiraling whitefly (Hemiptera: Aleyrodidae) in Florida. Florida Entomol. 2016;99(1):150–53. https://doi.org/10.1653/024.099.0134

98. Bhavani B. First report of rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin, an invasive pest on sugarcane in Andhra Pradesh, India. J Entomol Zool Stud. 2020;8(6):1993–99.

99. Poorani J, Thanigairaj R. First report of *Encarsia dispersa* Polaszek (Hymenoptera: Aphelinidae) as a parasitoid of rugose spiralling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae), a recent invasive pest in India, with notes on its predators. J Biol Control. 2017;31(1):1–4. https://doi.org/10.18311/jbc/2017/16263

100. Krishnarao G, Chalapathi Rao NB. Surveillance and eco-friendly management of new invasive alien pest, rugose spiralling whitefly, *Aleurodicus rugioperculatus* Martin: Inherent menace. J Appl Zool Res. 2019;30(2):148–58.

101. Abitha KR, Sushama V. First report of rugose spiraling whitefly, *Aleurodicus rugioperculatus* Martin (Hemiptera: Aleyrodidae) on wild date palm, *Phoenix sylvestris* (L.) Roxb. (Arecaceae), along with natural parasitization in West Bengal, India. Rec Zool Surv India. 2022:299–302. https://doi.org/10.26515/rzsi/v122/i3/2022/164118

102. Dubey AK. First record of three exotic whitefly pests (Hemiptera, Aleyrodidae) from Andaman and Nicobar Islands, India. Entomon. 2023;48(1):77–82. https://doi.org/10.33307/entomon.v48i1.846

103. Suriya S, Preetha G, Balakrishnan N, Sheela J. Host plants of invasive whiteflies-rugose spiralling whitefly, *Aleurodicus rugioperculatus* Martin and Bondar's nesting whitefly, *Paraleyrodes bondari* Peracchi. Indian J Ecol. 2024;51(4):883–90. https://doi.org/10.55362/IJE/2024/4325

104. Sankarganesh E, Kusal R. New report of neotropical invasive Bondar's nesting whitefly, *Paraleyrodes bondari* peracchi (Hemiptera: Aleyrodidae) from West Bengal, India. Insect Environ. 2021;24(4):555–58.

105. Iaccarino FM. Descrizione di *Paraleyrodes minei* n. sp.(Homoptera: Aleyrodidae), nuovo aleirodide degli agrumi in Siria. Bolletino del Laboratori di Entomol Agra Filippo Sylvestri. 1990;46:131–49.

106. Hidayat P, Bintoro D, Nurulalia L, Basri M. Species, host range and identification key of whiteflies of Bogor and surrounding area. Indonesia: Lampung University; 2018. https://doi.org/10.23960/j.hptt.218127-150

107. Krishnappa C, Dubey AK, Verma A, Mahapatro GK. Occurrence of exotic whitefly, *Paraleyrodes minei* Iaccarino (Hemiptera: Aleyrodidae) and other whitefly species on fruit crops in Maharashtra, India. 3 Biotech. 2021;11(6):264. https://doi.org/10.1007/s13205-021-02831-7

108. Longo S, Rapisarda C. Spread of *Paraleyrodes minei* Iaccarino (nesting whitefly) in Italian citrus groves. EPPO Bull. 2014;44 (3):529–33. https://doi.org/10.1111/epp.12146

109. Hidayat P, Anisa RP. Diversity and host ranges of whiteflies in Mekarsari fruit park, Bogor: a comprehensive study of 20 whitefly species across 56 fruit plant pecies. Andalasian Int J Entomol. 2024;2(1):8–14. https://doi.org/10.25077/aijent.2.1.8-14.2024

110. Sánchez-Flores ÓÁ, Myartseva SN, García-Martínez O, Ruíz-Cancino E. A new species of the genus *Encarsia*1, parasitoid of the whitefly *Aleurodicus rugioperculatus* Martin2 in Mexico. Southwest Entomol. 2017;42(3):701–06. https://doi.org/10.3958/059.042.0308

111. Delvare G, Genson G, Borowiec N, Etienne J, Karime ALA, Beaudoin-Ollivier. Description of *Eretmocerus cocois* sp. (Hymenoptera: Chalcidoidea), a parasitoid of *Aleurotrachelus atratus* (Hemiptera: Aleyrodidae) on the coconut palm. Zootaxa. 2008;1723(1):47–62. https://doi.org/ 10.11646/zootaxa.1723.1.3

Additional information

Peer review: Publisher thanks Sectional Editor and the other anonymous reviewers for their contribution to the peer review of this work.

Reprints & permissions information is available at https://horizonepublishing.com/journals/index.php/PST/open_access_policy

Publisher's Note: Horizon e-Publishing Group remains neutral with regard to jurisdictional claims in published maps and institutional affiliations.

Indexing: Plant Science Today, published by Horizon e-Publishing Group, is covered by Scopus, Web of Science, BIOSIS Previews, Clarivate Analytics, NAAS, UGC Care, etc

See https://horizonepublishing.com/journals/index.php/PST/indexing_abstracting

Publisher information: Plant Science Today is published by HORIZON e-Publishing Group with support from Empirion Publishers Private Limited, Thiruvananthapuram, India.