Volume 29 (1) March 2026
ISSN 0975-1963

Insect Environment

Quarterly Journal

Not-for-profit journal promoting Insect Science

Articles with DOI
NAAS Rating 3.53

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An *AtmaNirbhar* initiative by Indian entomologists for promoting Insect Science

Published by
Abraham Verghese Insect And Nature Trust (AVIAN Trust), Bengaluru, India

Supported by
International Phytosanitary Research & Services (IPRS), Bengaluru
Shree Nidhi Agrochemicals, Vijayapura, Devanahalli
Bengaluru Rural

Editorial Board

***Cover Page:**
***Aphis odinae* (formerly *Toxoptera odinae*)**
Photo by Dr. Deepak, S.
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Insect Environment

(Quarterly journal to popularize insect study and conservation) ISSN 0975-1963
NAAS Rating 3.53

The first issue of Insect Environment was published in 1996. The sole objective of Insect Environment is to popularize insect study through popular, semi-technical and technical research notes, extension notes for managing insect pests, photographs, short blogs and essays on all aspects of insects. The journal is published quarterly, in March, June, September, and December.

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Short popular insect notes, review essays, new records, profiles, tributes, and views are acceptable. There are no page charges; each article should preferably not exceed 500 words. Authors can refer to back volumes available on the website for writing style. Good photographs are encouraged. A special insect photo gallery "Insect Lens" is to encourage professional and amateur photographs of insects. These will be published in the quarterly *Insect Environment.* The blogs are for quick dissemination of insect "news". These will be uploaded within a month of submission and will be on the website. Blogs should be about a hundred words with one photograph, in simple English.

This journal is unique in that it encourages articles from students to amateurs to professionals! It is hosted in CABI (as full text) and ZooBank, Indian Abstracts, Research Gate, Google Scholar etc. ensuring global coverage. Scientific articles are assigned DOI.

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IE Blog Contents

Editorial

Insect science and the next decade of AI

Artificial Intelligence [AI] is all pervasive. As end-users find it 'friendly', many 'experts' are emerging on the horizon. It is the new buzz word for all fund-seekers! It is in my opinion a useful tool to the extent we know its merits and demerits. It is the former we need to harp on and certainly add our 'intelligence' adequately. In fact, in our edit office a light compliment is passed on to me: AV (that's me!) is better than AI when it comes to writing and the gamut of edit skills. Of course, I feel pampered and the real AI should not feel bad!!

I believe there will be tremendous positive progress in the coming years and decades with the growth of AI and AGI. My optimism comes partly from observing how small mobile devices transformed daily life, something I first began to appreciate while in Hawaii, mainland USA, and later among teenagers in Singapore and Japan. Even as early as between 1994 and 1999, I noticed their rapid adoption and usefulness especially abroad. This prompted me to get my own mobile in 1999 which at that time was pretty expensive as call received was also charged. At the time, many "prophets of doom" were warning that mobile towers and phones would destroy birds, bees, and even human brains. Yet, over the past three decades, these devices have proliferated into billions, and instead of harming fauna, it is now missiles and conflict zones, such as those radiating through the Gulf region, that pose far greater threats.

However, the future of AI in insect and pest management is shaping up to be transformative, blending precision science with sustainability. Here's what experts are envisioning:

Key Future Innovations

Automated Insect Identification: AI and machine learning can analyze images or sensor data to identify insect species quickly and accurately, reducing reliance on manual expertise. This speeds up monitoring and makes large-scale surveillance feasible.

Early Detection & Predictive Modeling: AI systems can forecast pest outbreaks by analyzing weather, crop growth, and insect behavior patterns. This allows farmers to act before infestations escalate, minimizing crop losses.

Precision/smart insect control: Instead of blanket pesticide use, AI can guide targeted interventions deploying biopesticides, pheromone traps, or minimal chemical use exactly where needed. This reduces environmental damage and costs.

Smart Monitoring Networks: Drones, IoT sensors, and satellite imagery integrated with AI could create real-time pest maps, helping farmers and policymakers coordinate responses across regions.

Challenges and Considerations may include data quality and quantity as all AI models need vast, diverse datasets of insect images and behaviours, which are still being perhaps generated. Accessibility as smallholder farmers may struggle with cost and infrastructure unless solutions are made affordable through liberal public intervention; this is especially true in India and south Asia.

Integrating AI tools with traditional knowledge and ecological approaches without replacing them; and this requires natural human intelligence!

Insect Environment has consistently prioritized field-based natural history studies, recognizing the central role of natural intelligence and direct observation in advancing entomological knowledge. Consequently, purely dry-lab or highly theoretical papers remain a lower priority for the journal. At the same time, this stance does not preclude the use of precision data analysis, particularly approaches based on machine learning (ML) that are capable of efficiently handling large datasets and complementing robust field-derived evidence.

The future of crop protection is shifting from reactive, pesticide-heavy approaches to proactive, knowledge-driven pest management, especially in large farms, plantations, and commercial horticulture. This transformation is guided by advances in monitoring, forecasting, and precision interventions. At the same time, it is reasonable to assume that smallholder farms will continue to rely on natural farming principles and traditional practices, integrating low-cost ecological methods as core components of their production systems as encouraged by the Government of India.

AI may be fast, but is limited to the 'intelligence' fed to it. For example, a yet to be described and unpublished insect species cannot be image-identified by AI. …… and AI admits it…!

Abraham Verghese
Editor-in-Chief

As we enter the 29th volume of Insect Environment, we, the editors, sincerely thank our well-wishers and all contributing authors for their continued support and valuable submissions. We extend special appreciation to our referee team for their prompt and efficient handling of manuscripts, which has greatly strengthened the journal's publication process. We are also encouraged by the growing engagement with our weekly blogs, contributed by entomologists from diverse regions national and global, which are now eagerly awaited by nearly 6,000 readers each week.

Research articles

DOI: 10.55278/WIMM9118

An Annotated Catalogue of Indian Zygaenidae (Zygaenoidea, Lepidoptera)

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Abstract

The present checklist presents the first comprehensive documentation of published literature and online databases on Indian Zygaenid moths, recording 141 species distributed across 52 genera of four subfamilies: Chalcosiinae (85 species), Procridinae (52 species), Callizygaeninae (2 species) and Zygaeninae (2 species). For each species, details regarding type localities, distribution, and host plant data are included.

Keywords: lepidoptera, burnet moths, taxonomy, distribution, host plants.

Introduction

The family Zygaenidae, often referred to as burnet and forester moths, constitutes a significant taxon within the order Lepidoptera. Although most zygaenid species inhabit tropical areas, they are also well-distributed in temperate zones. Globally, it is represented by five subfamilies: Callizygaeninae Alberti, 1954, Chalcosiinae Walker, 1865, Procridinae Boisduval, 1828, Zygaeninae Latreille, 1809, and Inouelinae Efetov & Tarmann, 2017. In India Inouelinae family is not been recorded. So, India is represented by 4 subfamilies. The family comprises over 1,000 species globally, and the total number of described species continues to grow each year (Efetov *et al*. 2021; Ulaşli & Can 2021). This family exhibits significant diversity in mimetic patterns, chemical defence systems, olfactory organs, reproductive strategies, host plant utilisation, and diapause biology, alongside a highly fragmented biogeographical distribution (Spalding *et al*. 2013; Hofmann and Tremewan 2017; Ulaşli & Can 2021). A notable feature of many Zygaenidae is their advanced chemical defence mechanism. Chalcosiinae and Zygaeninae moth larvae can emit hydrogen cyanide (HCN) by enzymatically breaking down cyanoglucosides (Niehuis *et al*. 2006).

This catalogue compiles data scattered across the above-mentioned platforms into a single source for easy access to the researchers. The compilation resulted in 141 species of Indian Zygaenidae from 52 genera in four subfamilies. This is the first comprehensive compilation of Zygaenidae from India. Although utmost care has been taken to ensure accuracy and precision, the possibility of inadvertent errors cannot be completely excluded.

Methodology

A thorough literature review was conducted, including primary sources such as scientific publications, academic journals and secondary sources like online databases and forums (Catalogue of Life; Savela, 2025). We have consulted the available literature by Efetov *et al*. 2012, Hofmann & Tremewan 2017, Efetov & Tarmann 2024.

Results and discussion

The abbreviation used in the Results section is DD (Data Deficient). Graphical representations are provided to facilitate interpretation of species and genera diversity within the family. The subfamily-wise species and genera counts are presented in Fig. 1, while the genus-wise species count is shown in Fig. 2. Fig. 3 illustrates the decade-wise discovery of species, Fig. 4 presents the total species count of Zygaenidae from India, and Fig. 5 depicts the research contributions during the pre-independence, post-independence, and recent periods. All graphs were prepared using Microsoft Excel.

As mentioned earlier this catalogue includes 141 species among which, Chalcosiinae comprises of 85 species in 34 genera, Procridinae 52 species within 16 genera, Callizygaeninae consists of two species from one genus, and Zygaeninae with two species in one genus (Figure 1). A total of 52 genera were documented from India, and the graphical representation shows that the genus *Artona* has the highest species count with 13 species; *Chrysartona* with 8 species, *Campylotes* and *Clelea* with 7 species (Figure 2).

Brief summaries regarding the key diagnostic and ecological importance for the four subfamilies of Zygaenidae recorded from India are provided below:

1. Chalcosiinae Walker, 1854, subfamily, is the most diverse in India, comprising large, brightly coloured and diurnal moths. A key feature is the presence of a specialised hindwing abdominal scent organ composed of a pleural fold between the first and second abdominal pleurites and a bundle of hair tufts arising from the anal axillary sclerite of the hindwing that fits into this fold, which plays a role in pheromone dissemination (Haase 1888; Tarmann 1992; Yen *et al*. 2005). Another remarkable characteristic is the ability to exude a large quantity of cyanogenic defensive secretion, often producing a hissing sound, released as foam through a dorsolateral opening located between the patagia and parapatagia; the precise storage site of this fluid remains unknown (Yen *et al*. 2005). The compound eyes lack interommatidial setae, a condition regarded as a secondary reduction within Zygaenidae (Yen 2003). The chaetosema is characterised by upright scales intermixed with sensilla trichodea, which is taxonomically informative. In the male genitalia, the tegumen bears specialised apodemes, while in females' abdominal segments VIII–X are modified into a well-developed ovipositor (Yen 2003). Additionally, the accessory gland on the utriculus of the receptaculum seminis is absent, providing another distinguishing reproductive character (Yen *et al*. 2005). These features collectively represent the most notable morphological and physiological traits used for identification of the subfamily. It represents the largest share with 85 species in 34 genera from India.

2. Procridinae Boisduval, 1828, subfamily, commonly known as "foresters", includes generally smaller moths that are metallic green, blue, or brassy in appearance. It exhibits several distinctive morphological, ultrastructural, and behavioural characters that are important for its identification within Zygaenidae. In females, the spermatheca (receptaculum seminis) is not differentiated into a bulb-like lagena and a tubular utriculus as in most ditrysian Lepidoptera; instead, it forms a long tubular structure, resembling the monotrysian condition. This is considered a secondary reduction or fusion of the lagena with the utriculus, and the slightly expanded portion is referred to as the bursa utricularis, a unique feature of Procridinae (Naumann 1988; Epstein *et al*. 1998; Kristensen 2003). Another notable trend is the enlargement of the posterior ductus bursae to form a praebursa, which can accommodate the spermatophore and partly replaces the function of the corpus bursae; the internal sclerotised structures often show species-specific variation and therefore have high diagnostic value (Alberti 1954; Tarmann 1984, 2004). Wing scales also show specialised ultrastructure, evolving from primitive transverse striae to advanced types such as plate-type, central-hole type, grid or sieve type, and ladder-type scales, while primitive scale patterns may still occur on certain body regions (Tarmann 1984, 2004; Kristensen & Simonsen 2003). Females display a characteristic calling behaviour using pheromone glands located on abdominal tergites III–V, exposing these segments by adopting a specific posture to release pheromones; the chemical composition of sex attractants in Procridinae consists mainly of esters of 2-butanol and fatty acids (Hallberg & Subchev 1997; Nishihara & Wipking 2003; Subchev *et al*. 2014). Additionally, the chaetosema shows a distinctive honeycomb-like arrangement in which upright scales surround the sensilla trichodea, providing another useful diagnostic character (Yen *et al*. 2005; Tarmann 2004). These combined reproductive, structural, and behavioural features represent key characters defining the subfamily Procridinae. It accounts for 52 species in 16 genera from India.

3. Zygaeninae Latreille, 1809, subfamily, commonly known as burnet moths, represent one of the most well-studied groups within Zygaenidae. Species are typically medium-sized with metallic blue or green forewings marked by red, orange, or yellow spots, which function as aposematic signals. Members of this subfamily known for their strong chemical defenses, containing cyanogenic glycosides that make them unpalatable to predators (Naumann *et al*. 1999). The compound eyes lack interommatidial setae, a condition regarded as a secondary reduction within Zygaenidae (Yen 2003). The chaetosema is situated on an elongated oval plate positioned dorsally or laterodorsally above the ocellus, appearing as a shallow triangular depression behind it; this structure consists of a central trichoid sensory area surrounded or bordered by upright scales, forming a distinctive arrangement useful for identification (Yen *et al*. 2005). In males, a well-developed abdominal coremata organ is typically present, consisting of a pair of eversible brush-like structures located on the intersegmental membrane between abdominal segments VIII and IX; however, this organ is secondarily reduced in certain taxa such as *Zygaena anthyllidis* and members of the *Z. loti* species group (Kames 1980; Efetov 2004, 2005). These characters collectively represent notable diagnostic traits of the subfamily. It accounts for 2 species in 1 genus from India.

4. Callizygaeninae Alberti, 1954, subfamily, is a relatively small and less diverse subfamily within Zygaenidae, containing few genera with restricted distribution mainly in parts of Asia and Africa. The group is considered morphologically primitive compared to other zygaenid subfamilies (Epstein *et al*. 1998). It possesses a combination of primitive and derived characters that help in its recognition within Zygaenidae. Among the plesiomorphic features, the medial stem in the wing venation is fully developed, the lagena is present in the receptaculum seminis, and Petersen's gland is absent, based on studies of *Callizygaena aurata* (Alberti 1954). In addition, several possible apomorphic characters have been identified. The compound eyes lack interommatidial setae, which is considered a secondary reduction within the family (Yen 2003). The foretibial epiphysis is reduced, and the bulla seminalis forms a globular bulb connected to the ductus seminalis through a narrow tubular stalk, although this condition requires further confirmation in additional species (Alberti 1954). The corpus bursae contain distinct signum plates, and the female abdominal segments VIII–X are modified to form a short ovipositor (Alberti 1954). It accounts for 2 species in 1 genus from India.

Looking upon the trend of species discovery in this family, Figure 3 shows that most descriptions were made during the late nineteenth century. The highest number of species (31) was described between 1890 and 1900. This was followed by 23 species during

1850–1860 and 21 species during 1870–1880. A moderate number of species were described in the periods 1840–1850 (14 species) and 1900–1910 (13 species). In recent years, the rate of discovery has been very low, with only six species reported between 2000 and 2010, two species between 2010 and 2020, and only one species from 2020 to 2026. Work on the description of the species was conducted during 1790–1830, 1930–1940, and 1960–2000, indicating long gaps in taxonomic work.

The contribution of different authors shows that Moore described the largest number of species (22), followed by Walker (19) and Hampson (18). Other researchers such as Elwes, Jordan, Swinhoe, Doubleday, and Butler also made important contributions. In recent years, only a few additions have been made, including one species described by Chada *et al.* (2017) and another by Sondhi *et al.* (2023), both from Arunachal Pradesh (Figure 4).

When the discoveries are grouped into pre-independence, post-independence, and recent periods, it becomes clear that most species (93%) were described before independence. Only 6% were described after independence, and recent discoveries account for just 1% (Figure 5). This suggests that although the family was studied extensively in earlier times, there is still a need for more surveys and taxonomic research to understand its diversity more completely. This catalogue is a significant step towards a comprehensive overview of the known Zygaenidae fauna of India.

Order LEPIDOPTERA LINNAEUS, 1758

Suborder GLOSSATA FABRICIUS, 1775

Infraorder HETERONEURA TILLYARD, 1918

Clade DITRYSIA BÖRNER, 1925

Superfamily ZYGAENOIDEA LATREILLE, 1809

Family ZYGAENIDAE LATREILLE, 1809

Subfamily **Callizygaeninae Alberti, 1954**

***Callizygaena* Felder, 1874**

Type species: *Callizygaena nivimacula* Felder; *Reise Fregatte Novara, Bd* 2 (Abth. 2) (4): pl. 83, f. 4.

1. *Callizygaena auratus* (Cramer, 1779) : 126, pl. 264, f. A (*Sphinx*)

= *Callizygaena auricincta* Swinhoe, 1897: 62; **Type Locality**: Nilgiri Hills

= *Tascia gana* Swinhoe, 1891: 133; **Type Locality**: Ganjam

Type Locality: Ceylon (Cramer, [1779])

Distribution: India: Tamil Nadu, Odisha; Sri Lanka (Swinhoe, 1891, 1892; Cramer, [1779])

Host Plant: DD

2. *Callizygaena aurifasciata* Hering, 1922:12

Type Locality: East India (Hering 1922)

Distribution: India: East India (Hering, 1922)

Host Plant: DD

Subfamily **Chalcosiinae Walker, 1865**

***Agalope* Walker, 1854**

Type species: *Agalope basalis* Walker; *List Spec. Lepid. Insects Colln Br. Mus*. 2: 437

3. *Agalope eronioides* (Moore, 1879):15 (*Chelura*)

Type Locality: Darjiling (Moore, 1879a)

Distribution: India: Assam, Meghalaya, West Bengal (Joshi *et al*., 2021; Shah *et al*., 2018)

Host Plant: DD

4. *Agalope hyalina* (Kollar, [1844]) : 462 (*Chalcosia*)

= *Agalope basalis* Walker, 1854: 438; **Type Locality**: N. India

Type Locality: in Himaleya, Massuri (Kollar, [1844])

Distribution: India: North India, Himachal Pradesh, Sikkim, Uttarakhand, Manipur, West Bengal; Afghanistan, Myanmar (Joshi *et al*., 2021; Pathania *et al*., 2021; Sondhi *et al*., 2026)

Host Plant: *Cotoneaster bacillaris*, *Malus pumila* (Rosaceae) (Robinson *et al*., 2010).

5. *Agalope livida* Moore, 1879: 391

= *Chalcosia davidi* Oberthür, 188

Type Locality: China (Moore, 1879)

Distribution: India: Assam, Sikkim (Sondhi *et al*., 2026)

Host Plant: DD

6. *Agalope primularis* Butler, 1875: 392

Type Locality: Darjeeling

Distribution: India: North India, West Bengal (Sondhi *et al*., 2026)

Host Plant: DD

***Paragalope* Huang, Horie, Fan, Wang and Espeland, 2023**

7. *Paragalope basiflava* (Moore, 1879): 391 (*Chelura*)

Type Locality: Darjeeling (Moore, 1879b)

Distribution: India: West Bengal (Shah *et al*., 2018)

Host Plant: DD

8. *Paragalope glacialis* (Moore, 1872): 570 (*Chelura*)

≡ *Agalope glacialis* (Moore, 1872): 26 (Ambiguous)

Type Locality: Darjiling (Moore, 1872)

Distribution: India: West Bengal (Sondhi *et al*., 2026)

Host Plant: DD

***Amesia* Duncan [and Westwood], 1841**

Type species: *Phalaena sanguiflua* Drury; *in Jardine, Naturalist's Libr*. (Edn. 1) 33 (Ent. 7): 93

9. *Amesia aliris* (Doubleday, 1847): 74 (*Gynautocera*)

Type Locality: Silhet (Doubleday, 1847)

Distribution: India: Assam, West Bengal (Shah *et al*., 2018; Joshi *et al*., 2021)

Host Plant: DD

10. *Amesia namouna* (Doubleday, 1847):74 (*Gynautocera*)

= *Erasmia desmiata* Jordan, 1907:25; **Type Locality**: W.Java, Mt Gede

= *Amesia modesta* Snellen, 1902: 178; **Type Locality**: Borneo

= *Amesia radiata* Walker, 1892:79

= *Amesia hyala* Druce, 1885: 518; **Type Locality**: Borneo; Darjeeling

= *Amesia pexifascia* Butler, 1877: 115; **Type Locality**: Malakka

= *Cyclosia semiradiata* Walker, 1865: 113; **Type Locality**: Sumatra

= *Cyclosia noctipennis* Walker, 1862:95

= *Epyrgis euploeoides* Herrich-Schäffer, 1853: 9-10

Type Locality: Northern India (Doubleday, 1847)

Distribution: India: Northern India (Doubleday, 1847)

Host Plant: DD

11. *Amesia sanguiflua* (Drury, 1773):34 (*Phalaena*)

= *Amesia sanguiflua korintjensis* Talbot, 1929:92 **Type Locality**: Sumatra, Mt Korintji

= *Amesia sanguiflua* Hering, 1922: 43 **Type Locality**: S.Formosa, Polisha

= *Erasmia javana* Dohrn, 1906:173

= *Erasmia lugens* Dohrn, 1906 173; **Type Locality**: Sumatra

= *Amesia sanguiflua gedeana* Fruhstorfer, 1897: 43;

Type Locality: S.Formosa, Polisha

Type Locality: India: Sikhim, Khasis, Shillong (Drury, 1773)

Distribution: India: Meghalaya, West Bengal (Shah *et al*., 2018; Joshi *et al*., 2021)

Host Plant: DD

***Arbudas* Moore, 1879**

Type species: *Arbudas bicolor* Moore; *Descr. Indian lep. Atkinson* (1): 19

12. *Arbudas bicolor* Moore, 1879: 20

Type Locality: Darjiling (Moore, 1879a)

Distribution: India: Assam, Meghalaya (Joshi *et al*., 2021); Vietnam (Moore, 1879)

Host Plant: DD

13. *Arbudas truncatus* (Jordan, 1907): 36 (*Pidorus*)

Type Locality: Sikkim (Jordan, 1907)

Distribution: India: Sikkim; Myanmar (Burma) (Yen *et al*., 2005)

Host Plant: DD

14. *Arbudas leno* (Swinhoe, 1900): 305 (*Pidorus*)

Type Locality: Jaintia Hills (Swinhoe, 1900)

Distribution: India: Assam, Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

***Pseudarbudas* Tarmann, 1992**

Type species: *Cyclosia ochrea* Elwes; *Heteroc. Sumatrana* 7: 45

15. *Pseudarbudas ochrea* (Elwes, 1890): 385 (*Cyclosia*)

≡ *Arbudas ochrea* (Elwes, 1890):13

Type Locality: Assam, Naga Hills (Elwes, 1890)

Distribution: India: Nagaland (Joshi *et al*., 2021)

Host Plant: DD

16. *Pseudarbudas efetovi* Tarmann, 2012:128

Type Locality: India, Khasia Hills (Tarmann, 2012)

Distribution: India: Meghalaya (Tarmann, 2012)

Host Plant: DD

***Boradia* Moore, 1879**

Type species: *Boradia carneola* Moore; *Proc. Zool. Soc. Lond*. 1879: 391

17. *Boradia carneola* Moore, 1879: 392

Type Locality: NW. Himalayas, Dharmsala (Moore, 1879b)

Distribution: India: Himachal Pradesh (Pathania *et al*., 2021)

Host Plant: DD

***Cadphises* Moore, [1866]**

Cadphises maculata Moore; *Proc. zool. Soc. Lond*. 1865 (3): 800

18. *Cadphises maculata* Moore, [1866]: 801

Type Locality: Darjeeling (Moore, [1866])

Distribution: India: Assam, Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

19. *Cadphises moorei* Butler, 1875: 392

Type Locality: Darjeeling (Butler, 1875)

Distribution: India: Assam, Meghalaya, Nagaland, West Bengal (Joshi *et al*., 2021)

Host Plant: DD

***Campylotes* Westwood, 1839**

Type species: *Campylotes histrionicus* Westwood; *in Royle, III. Nat. Himalaya Mnt*. 11 Lep.: 53.

20. *Campylotes atkinsoni* Moore, 1879:17

Type Locality: Darjiling (Moore, 1879a)

Distribution: India: West Bengal (Shah *et al*., 2018)

Host Plant: DD

21. *Campylotes desgodinsi* (Oberthür, 1884): 18 (*Epyrgis*)

Type Locality: Tibet (Oberthür, 1884)

Distribution: India: Arunachal Pradesh, Nagaland, Manipur **(**Joshi *et al*., 2021)

Host Plant: DD

22. *Campylotes histrionicus* Westwood, 1839: 53

= *Campylotes excelsa* Oberthür, 1896: 178

= *Campylotes altissima* Elwes, 1890: 384

Type Locality: India: Himalays, Assam Hills (Westwood, 1839)

Distribution: India: Himachal Pradesh, Jammu and Kashmir, Meghalaya, Sikkim, Uttarakhand, West Bengal (Shah *et al*., 2018; Joshi *et al*., 2021); Afghanistan, China (Patania *et al*., 2021)

Host Plant: *Lyonia ovalifolia*, *Rhododendron* sp. (Ericaceae) (Robinson *et al.,* 2010)

23. *Campylotes kotzschi* Röber, 1926 :32

Type Locality: Naga-Hills in Assam (Röber, 1926)

Distribution: India: North India (Yen *et al*., 2005)

Host Plant: DD

24. *Campylotes sikkimensis* Elwes, 1890: 38

Type Locality: India: West Bengal (Shah *et al*., 2018)

Distribution: India: Assam, Meghalaya, North India (Jordan, 1907)

Host Plant: DD

25. *Campylotes desgodinsi splendida* Elwes, 1890: 384

≡ *Campylotes splendida* Elwes, 1890: 288

Type Locality: DD

Distribution: India: North India; Yunan and China (Elwes, 1890)

Host Plant: DD

26. *Campylotes wernickei* Röber, 1925: 170

Type Locality: Assam

Distribution: India: Himalaya, Northeast India; China, Myanmar, possibly nearby Asian regions (Sondhi *et al*., 2026)

Host Plant: DD

***Chalcosia* Hübner, [1819]**

Type species: *Sphinx pectinicornis* Linnaeus; *Verz. bek. Schmett*. (11): 173

27. *Chalcosia affinis* (Guérin-Méneville, 1843): 86 (*Gynautocera*)

Type Locality: Pondichery and Malayan coast **(**Guérin-Méneville, 1843)

Distribution: India: South India **(**Guérin-Méneville, 1843)

Host Plant: DD

28. *Chalcosia ideaoides* Herrich-Schäffer, [1853]: pl. 1

Type Locality: N. India (Yen *et al*., 2005).

Distribution: India: (Assam, NW. Himalayas); Nepal (Thapa, 1998).

Host Plant: DD

Remark. This species is synonymised under *Chalcosia pectinicornis ideaoides* Herrich-Schäffer, 1853 (Catalogue of Life 2026) but some literature shows it as *C. ideaoides*.

29. *Chalcosia pectinicornis* (Linnaeus, 1758): 495 (*Sphinx*)

= *Chalcosia formosicola* Matsumura, 1931: 285

= *Chalcosia guerini* Kirby, 1892: 45

= *Epyris virginalis* Ménétriés, 1857:143

= *Bombyx tiberina* Cramer, 1775:52

= *Papilio thallo* Linnaeus, 1767:756

= *Sphinx auxo* Linnaeus, 1767: 805

Type Locality: Afia (Linnaeus 1758)

Distribution: India: Assam, Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

30. *Chalcosia venosa* Walker, 1854: 422

= *Chalcosia myrrhina* Hampson, 1892: 265

= *Chalcosia thallo* Hampson, 1892: 266

= *Chalcosia quadrifasciata* Moore, 1882: 45

= *Chalcosia similata* Moore, 1882: 45

Type Locality: Ceylon (Walker, 1854)

Distribution: India: Himachal Pradesh; Sri Lanka **(**Pathania *et al.* 2021**)**

Host Plant: DD

***Chalcophaedra* Jordan, 1907**

Type species: *Gynautocera zuleika* Doubleday; *Gross-Schmett. Erde* 10: 39

31. *Chalcophaedra Zuleika* (Doubleday, 1847): 76 (*Gynautocera*)

= *Chalcosia corusca* Herrich-Schäffer, 1853:1

Type Locality: Silhet (Doubleday, 1847)

Distribution: India; Nepal and Bhutan (Sondhi *et al*., 2026)

Host Plant: DD

***Achelura* Kirby, 1892**

Type species: *Chelura bifasciata* Hope; *Syn. Cat. Lepid. Het*. 1: 56

32. *Achelura bifasciata* (Hope, 1841): 444 (*Chelura*)

Type Locality: Assam (Hope, 1841)

Distribution: India: (Northern parts, Uttarakhand, Sikkim, Meghalaya, Assam); Nepal, Bhutan, central Myanmar (Smith, 2010; Singh et al., 2023)

Host Plant: *Pyracantha crenulata* (Rosaceae) (Sondhi *et al*., 2021)

***Chalcosiopsis* Swinhoe, 1894**

Type species: *Chalcosiopsis variata* Swinhoe; *Ann. Mag. nat. Hist*. (6) 14 (84): 442

33. *Chalcosiopsis variata* Swinhoe, 1894: 442

Type Locality: Shillong (Swinhoe, 1894)

Distribution: India: Assam, Arunachal Pradesh, Meghalaya (Joshi *et al*., 2021

Host Plant: DD

***Corma* Walker, [1865]**

Type species: *Corma obscurata* Walker; *List Spec. Lepid. Insects Colln Br. Mus*.: 31

34. *Corma maculata* Hampson, [1893]: 268

Type Locality: Chin Hills, W. Burma (Hampson, [1893])

Distribution: India: Assam, Arunachal Pradesh, Meghalaya, West Bengal, Nagaland (Sondhi *et al*., 2026)

Host Plant: DD

35. *Corma zelica* (Doubleday, 1847): 76 (*Gynautocera*)

=*Codane neoterica* Swinhoe, 1890: 177

= *Codane leucomelas* Moore, 1886: 97; **Type Locality**: Ponsokai, Siam

Type Locality: Silhet (Doubleday, 1847)

Distribution: India: Assam; Bangladesh (Sondhi *et al*., 2026)

Host Plant: DD

36. *Corma zenotia* (Doubleday, 1847): 77 (*Gynautocera*)

= *Pidorus zenotea* Walker, 1854: 425

= *Corma spoliata* Walker, 1865: 125

Type Locality: Silhet (Doubleday, 1847)

Distribution: India: Assam, Meghalaya (Joshi *et al*., 2021) Bangladesh (Sondhi *et al*. 2026)

Host Plant: DD

***Cyclosia* Hübner, [1820]**

Type species: *Phalaena panthona* Stoll; *Verz. bek. Schmett*. (12): 177

37. *Cyclosia imitans* (Butler, 1881): 24 (*Epyrgis*)

Type Locality: Bhutan {Lidderdale) (Butler, 1881)

Distribution: India: Assam, Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

38. *Cyclosia inornata* (Walker, [1865]): 111 (*Pompelon*)

= *Mimeuploea tristis* Jordan, 1902: 274; **Type Locality**: North Borneo, Sandakan

= *Epyrgis cuprea* Swinhoe, 1891: 475; **Type Locality**: Khasia Hills

Type Locality: Sumatra (Walker, [1865])

Distribution: India: Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

39. *Cyclosia latipennis* (Hampson, 1891): 45 (*Pintia*)

Type Locality: Nilgiris (Hampson, 1891)

Type depository: BMNH (Hampson, 1891)

Distribution: India: Kerala, Karnataka, Goa, Maharashtra (Sondhi *et al*., 2026)

Host Plant: DD

40. *Cyclosia midama* (Herrich-Schäffer, [1853]): 78 (*Epyrgis*)

= *Cyclosia latistriga* Talbot, 1929: 92; **Type Locality**: Sumatra

= *Isbarta extrema* Grünberg, 1908: 289

= *Isbarta maassi* Grünberg, 1908: 288; **Type Locality**: SE.Borneo; Labuan

= *Callamesia hormenia* Hampson, 1892

= *Amesia striata* Druce, 1891: 86; **Type Locality**: Borneo, Labua

= *Amesia stelligera* Butler, 1881: 23; **Type Locality**: Bhutan

= *Amesia juvenis* Butler, 1879: 559; **Type Locality**: Malacca

= *Cyclosia spilophila* Walker, 1869: 4

= *Cyclosia venusta* Walker, 1854: 434; **Type Locality**: Silhet

= *Epyrgis hormenia* Herrich-Schäffer, 1853: pl. 2, f. 8; TL: Ceylon

Type Locality: Ind. bor. (Herrich-Schäffer, [1853])

Distribution: India: Assam, Meghalaya, Nagaland (Joshi *et al.* 2021)

Host Plant: DD

41. *Cyclosia panthona* (Stoll, [1780]) : 68 (*Geometra*)

Type Locality: China (Stoll, [1780])

Distribution: India: Assam, Bihar, Meghalaya, Sikkim (Joshi *et al.,* 2021); China, Hong Kong, Sri Lanka, Malaysia, Myanmar, Thailand (Beccaloni, 2003; Hampson, 1892)

Host Plant: *Porosa cardiosperma*, *Aporosa villosa*, *Baccaurea ramiflora* (Phyllanthaceae), *Hydnocarpu*s (Achariaceae) (Robinson *et al*., 2010)

42. *Cyclosia papilionaris* (Drury, 1773): 4 (*Noctua*)

= *Cyclosia obsoleta* Dufrane, 1936: 122; **Type Locality**: Tonkin

= *Cyclosia pallida* Dufrane, 1936: 122; **Type Locality**: Tonkin

= *Cyclosia enodis* Swinhoe, 1892: 69

= *Pintia latipennis* Hampson, 1891: 45

= *Epyrgis parvula* Butler, 1883: 160

= *Eterusia ferrea* Walker, 1854: 431

Type Locality: China (Drury). 'In Indis" (Fabr.) (Drury, 1773)

Distribution: India: Andaman and Nicobar Islands; Thailand, southern China (Sondhi *et al*., 2026)

Host Plant: *Dipterocarpus tuberculatus* (Dipterocarpaceae) (Robinson *et al*., 2010)

***Elcysma* Butler, 1881**

Type species: *Elcysma translucida* Butler; *Trans. R. ent. Soc. Lond.*: 4

43. *Elcysma dohertyi* Elwes, 1890: 386

Elcysma westwoodi dohertyi Elwes, 1890: 26

Type Locality: Naga Hills (Elwes, 1890)

Distribution: India: Assam, Manipur, Nagaland (Joshi *et al.*, 2021)

Host Plant: DD

44. *Elcysma ziroensis* Chada, Gogoi and Young, 2017: 11061

Type Locality: Talle Wildlife Sanctuary, Zrio, Arunachal Pradesh (Chada *et al*., 2017)

Distribution: India: Arunachal Pradesh (Chada *et al*., 2017)

Host Plant: *A. Prunus* species (Chada *et al*., 2017)

***Erasmia* Hope, 1841**

Type species: *Erasmia pulchella* Hope; *Trans. linn. Soc. Lond*. 18: 446

45. *Erasmia pulchella* Hope, 1841: 446

= *Erasmia chinensis* Hering, 1922: 12

= *Erasmia conjuncta* Strand, 1916: 147; **Type Locality**: Formosa

= *Erasmia hainana* Jordan, 1909: 24; **Type Locality**: Hainan

= *Erasmia cyanea* Jordan, 1907: 24; **Type Locality**: Hongkong

Type Locality: Assam (Hope, 1841)

Distribution: India: Assam, Meghalaya, Nagaland (Joshi *et al*., 2021)

Host Plant: DD

***Eterusia* Hope, 1841**

Type species: *Eterusia tricolor* Hope; *Trans. linn. Soc. Lond*. 18: 445

46. *Eterusia aedea* (Linnaeus, 1763): 403 (*Papilio*)

= *Eterusia okinawana* Matsumura, 1927: 83; **Type Locality**: Okinawa

= *Eterusia sakaguchii* Matsumura, 1927: 83; **Type Locality**: Okinawa

= *Eterusia postlutea* Strand, 1916: 142

= *Eterusia lepcha* Jordan, 1907: 34; **Type Locality**: Burma

= *Heterus dulcis* Butler, 1881: 21; **Type Locality**: Darjiling

= *Eterusia magnifica* Butler, 1879: 5; **Type Locality**: Cachar

= *Heterusia signata* Möschler, 1872: 341

= *Eterusia sinica* Ménétriés, 1857: 144; **Type Locality**: China

Type Locality: India (Linnaeus, 1763)

Distribution: India: Sikkim, Assam, Meghalaya, Nagaland; China, Nepal, Bangladesh, Sri Lanka, Thailand, Japan. (Kendrick *et al*., 2011)

Host Plant: *Buddleja* sp. (Scrophulariaceae), *Camellia sinensis* (Theaceae) (Robinson *et al*., 2010)

47. *Eterusia raja* Moore, 1859: 320

Type Locality: Darjeling (Moore, 1859)

Distribution: India: Assam, Arunachal Pradesh (Joshi *et al*., 2021; Sondhi *et al*., 2026)

Host Plant: DD

48. *Eterusia repleta* Walker, [1865]: 118

= *Eterusia alompra* Hampson, 1892: 16; **Type Locality**: Sibsagar, Assam

= *Eterusia urania* Schaus, 1890: 39; **Type Locality**: Naga Hills, Assam

Type Locality: Cambodia (Walker, [1865])

Distribution: India: Assam, Sikkim, Meghalaya, Nagaland, Odisha; Myanmar (Joshi *et al*., 2021; Sondhi *et al.*, 2026)

Host Plant: DD

49. *Eterusia sublutea* Walker, 1854: 430

= *Heterusia scintillans* Herrich-Schäffer, 1854: f. 154-155; **Type Locality**: India

Type Locality: Silhet (Walker, 1854)

Distribution: India; Bangladesh, Myanmar, Sylhet (Sondhi *et al*., 2026)

Host Plant: DD

50. *Eterusia tricolor* Hope, 1841: 445

= *Heterusia trimacula* Möschler, 1872:342

Type Locality: Assam (Hope 1841)

Distribution: India: Assam; Myanmar (Sondhi *et al.*, 2026)

Host Plant: DD

***Eumorphiopais* Hering, 1922**

Type species: *Eumorphiopais quadriplaga* Hering; *Archiv f. Naturg*. 88 A (11): 17

51. *Eumorphiopais leis* (Swinhoe, 1894): 442 (*Heteropan*)

Type Locality: Shillong (Swinhoe 1894)

Distribution: India: Assam; Bangladesh Thailand, laos (Tarmann, 1992)

Host Plant: DD

Gynautocera **Guérin-Méneville, 1831**

Type species: *Gynautocera papilionaria* Guérin-Méneville *Mag. Zool. Paris (Ins.)* 1: 12

52. *Gynautocera papilionaria* Guérin-Méneville, 1831: 12

Gynautocera papilionaria Guérin-Méneville, 1831; *Mag. Zool. Paris (Ins.)* 1: 12

= *Chalcosia selene* Kollar, 1894: 463; **Type Locality**: Caschmir

= *Gynautocera rara* Hampson, 1892: 279

= *Gynautocera zara* Swinhoe, 1891: 21

= *Histia fraterna* Moore, 1883: 15

Type Locality: DD

Distribution: India: Assam, Jharkhand, Himachal Pradesh, Jammu and Kashmir, Maharashtra, Meghalaya, Nagaland, Sikkim, Uttarakhand, Uttar Pradesh and West Bengal; Bangladesh, Bhutan, Myanmar, Nepal, China, Sri Lanka, Vietnam (Pathania *et al*. 2021)

Host Plant: *Litsea monopetala* (Lauraceae) (Smetacek *et al*., 2011)

Neoherpa **Tremewan, 1973**

Type species: *Herpa venosa* Walker *Bull. Br. Mus. nat*. (*Ent.*) 28 (3): 134

53. *Neoherpa primulina* (Elwes, 1890): 382 (*Herpa*)

Type Locality: Naga Hills (Elwes 1890)

Distribution: India: North East (Sondhi *et al*., 2026)

Host Plant: DD

54. *Neoherpa subhyalina* (Moore, 1879): 18 (*Herpa*)

= *Herpa elongatissima* Oberthür, 1910: 314; **Type Locality**: Tse-kou

Type Locality: Lachung Valley, Sikhim (Moore 1879a)

Distribution: India: North East (Sondhi *et al*., 2026)

Host Plant: DD

55. *Neoherpa venosa* (Walker, 1854) : 442 (*Herpa*)

Type Locality: Silhet (Walker 1854)

Distribution: India: North East (Sondhi *et al*., 2026)

Host Plant: DD

Hampsonia **Swinhoe, 1894**

Type species: *Hampsonia pulcherrima* Swinhoe; *Ann. Mag. nat. Hist*. (6) 14 (84): 443

56. *Hampsonia pulcherrima* Swinhoe, 1894: 443

Type Locality: Cherra Punji; Khasi Hills (Swinhoe, 1894)

Distribution: India: Meghalaya (Swinhoe, 1894)

Host Plant: DD

***Herpidia* Bryk, 1936**

Type species: *Herpa eupoma* Swinhoe 1897; *Ann. Mag. nat. Hist*. 19: 166

57. *Herpidia eupoma* (Swinhoe, 1897): 166 (*Herpa*)

Type Locality: Jaintia Hills (Swinhoe, 1897)

Distribution: India: Meghalaya (Joshi *et al.,* 2021)

Host Plant: DD

***Heteropan* Walker, 1854**

Type species: *Heteropan scintillans* Walker *List Spec. Lepid. Insects Colln Br. Mus*. 2: 440

58. *Heteropan scintillans* Walker, 1854: 441

Type Locality: Ceylon (Walker, 1854)

Distribution: India: Sikkim, Uttarakhand; Sri Lanka (Hampson, 1892)

Host Plant: DD

***Heterusinula* Bryk, 1936**

Type species: *Eterusia dichroa* Jordan; *Lepid. Cat*. 71: 231

59. *Heterusinula dichroa* (Jordan, 1907): 33 (*Eterusia*)

Type Locality: Assam, Khasia Hills

Distribution: India: Assam (Jordan, 1907)

Host Plant: DD

***Histia* Hübner, [1820]**

Type species: *Zygaena flabellicornis* Fabricius *Verz. bek. Schmett*. (13): 198

60. *Histia flabellicornis* (Fabricius, 1775): 831 (*Zygaena*)

= *Histia nigrina* Jordan, 1907: 13

= *Histia albimacula* Hampson, 1892: 280; **Type Locality**: Momeit, Burma, 2000ft

= *Papilio rhodope* Cramer, 1775: pl. 30, f. E

Type Locality: America meridionali (Fabricius, 1775)

Distribution: India**:** Assam, Himachal Pradesh**,** Uttarakhand, Nagaland, Arunachal Pradesh, Bhutan (Pathania *et al*., 2021; Sondhi *et al*., 2026)

Host Plant: *Bischofia javanica* (Phyllanthaceae) (Robinson *et al*. 2010)

61. *Histia libelluloides* (Herrich-Schäffer, [1853]): 7 (*Gynautocera*)

= *Histia anobia* Dohrn, 1899: 252; **Type Locality**: Sumatra

= *Histia nivosa* Rothschild, 1896: 56; **Type Locality**: Kina Balu

= *Histia selene* Walker, 1854: 413

= *Histia vacillans* Walker, 1854: 413

Type Locality: Java

Distribution: India: Assam, Andaman and Nicobar Islands (Hampson 1892)

Host Plant: DD

***Milleriana* Herrich-Schäffer, 1856**

Type species: *Milleria virginalis* Herrich-Schäffer; *Samml. aussereurop. Schmett*. (I) 1: 78, 1 (1): pl. 1, f. 4

62. *Milleriana adalifa* (Doubleday, 1847): 76 (*Gynautocera*)

= *Chalcosia nitida* Jordan, 1907: 37; **Type Locality**: W. Sumatra

= *Milleria cyanivena* Hampson, 1892: 263

= *Epyrgis forbesi* Druce, 1882: 779; **Type Locality**: Java, Bantam

= *Cyclosia cardinalis* Moore, 1879: 18; **Type Locality**: Sikkim

= *Cyclosia fuliginosa* Walker, 1854: 418

= *Cyclosia subcyanescens* Walker, 1854: 417

= *Chalcosia virginalis* Herrich-Schäffer, 1853: 78

= *Gynautocera adalifa* Doubleday, 1847

Type Locality: N. India

Distribution: India: Meghalaya, Uttarakhand (Sondhi *et al*., 2026)

Host Plant: DD

63. *Milleriana dualis* (Hering, 1941): 82 (*Milleria*)

Type Locality: N. India (Hering, 1941)

Distribution: India: North India (Savela, 2026)

Host Plant: DD

64. *Milleriana hamiltoni* (Swinhoe, 1891): 475 (*Milleria*)

Type Locality: Khasi Hills, Assam **(**Swinhoe, 1891)

Distribution: India: Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

***Philopator* Moore, [1866]**

Type species: *Philopator basimaculata* Moore 1865; *Proc. zool. Soc. Lond*.: 800, pl.42, fig.6

65. *Philopator basimaculata* Moore, [1866]: 800

Type Locality: Sikkim, Darjeeling (Moore, [1866])

Distribution: India: Sikkim, Meghalaya, Nagaland, Manipur (Thapa 1998)

Host Plant: DD

66. *Philopator rotunda* Hampson, 1896: 470

Type Locality: Sikkim; Khasis; Nagas; Manipur (Hampson, 1896)

Distribution: India: Manipur, Meghalaya, Nagaland (Joshi *et al*., 2021)

Host Plant: DD

***Phlebohecta* Hampson, [1893]**

Type species: *Soritia fuscescens* Moore; *Fauna Br. India (Moths)* 1: 251

67. *Phlebohecta fuscescens* (Moore, 1879): 16 (*Soritia*)

Type Locality: Darjiling (Moore, 1879a)

Distribution: India: Assam, Meghalaya (Joshi *et al.*, 2021)

Host Plant: DD

68. *Phlebohecta lithosina* (Felder, 1874): 83 (*Paraphlebia*)

= *Retina ? flavicosta* Elwes, 1890: 386; **Type Locality**: Sikkim

= *Soritia lithosia* Oberthür, 1923: 210

Type Locality: Borneo (Felder, 1874)

Type depository: Not known.

Distribution: India: Sikkim; Myanmar (Bhattacharya, 2003)

Host Plant: DD

***Pidorus* Walker, 1854**

Type species: *Phalaena glaucopis* Drury; *List Spec. Lepid. Insects Colln Br. Mus*. 2: 424

69. *Pidorus albifascia* (Moore, 1879): 19 (*Milleria*)

Type Locality: Cherra Punji, Assam, India (Moore, 1879a)

Distribution: India: Assam, Meghalaya, Nagaland (Joshi *et al*., 2021)

Host Plant: DD

70. *Pidorus circe* (Herrich-Schäffer, [1853]) : pl.1 (*Chalcosia)*

= *Laurion metallica* Walker, 1854: 426; **Type Locality**: N. India

Type Locality: Silhet **(**Herrich-Schäffer, [1853])

Distribution: India: Assam, Manipur, Meghalaya, Nagaland (Joshi *et al*., 2021)

Host Plant: DD

71. *Pidorus circinata* (Herrich-Schäffer, [1854]): 79 (*Heterusia*)

= *Eterusia fasciata* Walker, 1869: 5; **Type Locality**: Sikkim

Type Locality: Assam (Herrich-Schäffer, [1854])

Distribution: India: Assam, Meghalaya (Joshi *et al.*, 2021)

Host Plant: DD

72. *Pidorus gemina* (Walker, 1854): 427 (*Laurion)*

Type Locality: India: North; Hong Kong, North China, East Indies, Silhet (Walker 1854)

Distribution: India: Meghalaya (Joshi *et al*., 2021)

**Host Plant*:* *Aporosa villosa* (Savela,2026)

73. *Pidorus glaucopis* (Drury, 1773): 11 (*Phalaena)*

= *Bombyx rubrocollaris* Goeze, 1781

Type Locality: Bengal (Drury 1773)

Distribution: India: Assam, Meghalaya, Nagaland (Joshi *et al*., 2021)

Host Plant: DD

74. *Pidorus miles* (Butler, 1881): 25 (*Laurion*)

Type Locality: Darjiling [Lidderdale) (Butler, 1881)

Distribution: India: West Bengal (Shah *et al*., 2018)

Host Plant: DD

***Psaphis* Walker, 1854**

Type species: *Psaphis camadeva* Walker; *List Spec. Lepid. Insects Colln Br. Mus*. 2: 433

75. *Psaphis euschemoides* (Moore, [1866]): 802 (*Canerkes*)

= *Canerkes resumpta* Dohrn, 1906: 179

= *Canerkes javana* Röber, 1897: 7; **Type Locality**: W.Java

Type Locality: Cherra Poonjee; Silhet (Moore, [1866])

Distribution: India: Assam (Joshi *et al*., 2021)

Host Plant: DD

***Pseudoscaptesyle* Hering, 1922**

Type species: *Eterusia circumdata* Walker; *Archiv f. Naturg*. 88 A (11): 76

76. *Pseudoscaptesyle circumdata* (Walker, [1865]): 121 (*Eterusia*)

= *Pseudoscaptesyle citrana* Hering, 1922: 77; **Type Locality**: Sumatra

Type locality: Hindostan (India) (Walker [1865])

Distribution: India: Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

***Retina* Walker, 1854**

Type species: *Retina rubrivitta* Walker; *List Spec. Lepid. Insects Colln Br. Mus*. 2: 438

77. *Retina rubrivitta* Walker, 1854: 439

Type Locality: Silhet (Walker, 1854)

Distribution: India: Assam, Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

***Soritia* Walker, 1854**

Type species: *Chalcosia leptalina* Kollar; *List Spec. Lepid. Insects Colln Br. Mus*. 2: 435

78. *Soritia bicolor* (Moore, 1884): 355 (*Devanica*)

= *Soritia viridivena* Hampson, 1892: 253

Type Locality: Cachar (Moore, 1884)

Distribution: India: Assam, Meghalaya, Nagaland (Joshi *et al.*, 2021)

Host Plant: DD

79. *Soritia nigribasalis* Hampson, [1893]: 253

Type locality: Sikkim (Hampson, [1893])

Distribution: India: Meghalaya (Joshi *et al.*, 2021)

Host Plant: DD

80. *Soritia pulchella* (Kollar, [1844])

Chalcosia pulchella Kollar, [1844]; *in Hügel, Kaschmir und das Reich der Siek* 4: 461

= *Eterusia unipunctata* Dufrane, 1936: 122

= *Soritia xanthophlebia* Hering, 1922: 58; **Type Locality**: Khasia Hills

= *Soritia cicada* Felder, C. & Felder, R., 1874: 5

= *Heterusia flavomaculata* Möschler, 1872: 343; **Type Locality**: Silhet

= *Heterusia octopunctata* Möschler, 1872: 344

= *Eterusia triliturata* Walker, 1864: 119; **Type Locality**: Mussoorie

= *Gynautocera sexpunctata* Doubleday, 1847: 77

= *Chalcosia leptalina* Kollar, 1844: 462; **Type Locality**: Massuri

Type Locality: Himalaya, Massuri (Kollar, [1844])

Distribution: India: Meghalaya (Joshi *et al*., 2021)

Host Plant: *Camellia sinensis* (Theaceae), *Lagerstroemia indica* (Lythraceae), *Malus pumila*, *Rubus macilentus* (Rosaceae), *Melastoma malabathricum* (Melastomataceae) (Robinson *et al*., 2010)

81. *Soritia shahama* (Moore, [1866]): 801 (*Eterusia*)

Type Locality: Darjeeling (Moore, [1866])

Distribution: India: West Bengal (Shah *et al*., 2018)

Host Plant: DD

82. *Soritia sevastopuloi* Tremewan, 1959: 256

Type Locality: Assam, Shillong (Tremewan, 1959)

Distribution: India: Assam, Meghalaya (Tremewan, 1959)

Host Plant: DD

***Trypanophora* Kollar, [1844]**

Type species: *Trypanophora semihyalina* Kollarin Hügel, *Kaschmir und das Reich der Siek* 4: 457

83. *Trypanophora australis* Jordan, 1907: 15

Type Locality: Merkara, South India (Jordan, 1907)

Distribution: India: South India (Sondhi *et al*., 2026)

Host Plant: DD

84. *Trypanophora flavalis* Hampson, [1893]: 250

Type Locality: Matelei, Burma (Hampson, [1893])

Distribution: India; Myanmar (Sondhi *et al*., 2026)

Host Plant: DD

85. *Trypanophora hosemanni* Hering, 1922: 20

Type locality: Darjeeling? (Hering, 1922)

Distribution: India: West Bengal (Sondhi *et al*., 2026)

Host Plant: DD

86. *Trypanophora semihyalina* Kollar, [1844]: 457

= *Trypanophora atkinsoni* Moore, 1879: 15; **Type Locality**: Darjiling

= *Syntomis glaucopoides* Walker, 1864: 69

= *Syntomis humeralis* Walker, 1856: 1503

Type Locality: Kashmir (Kollar, [1844])

Distribution: India: Assam, Himachal Pradesh, Jammu and Kashmir, Meghalaya, Sikkim, Tripura, Uttarakhand (Joshi *et al*., 2021; Pathania *et al*., 2021)

Host Plant: *Arringtonia acutangula* (Lecythidaceae), *Carissa carandas* (Apocynaceae), *Lagerstroemia indica*, *Lagerstroemia speciosa* (Lythraceae), *Ricinus communis* (Euphorbiaceae), *Shorea robusta* (Dipterocarpaceae), *Terminalia catappa*, *Terminalia tomentosa* (Combretaceae), *Bombax ceiba* (Malvaceae), *Ziziphus jujuba* (Rhamnaceae) (Robinson *et al.*, 2010)

87. *Trypanophora trapobanes* Walker, 1854: 435

= *Trypanophora taprobanes* Moore, 1882: 40

Type Locality: Ceylon (Walker, 1854)

Distribution: India; Sri Lanka (Sondhi *et al*., 2026)

Host Plant: DD

Subfamily **Procridinae Boisduval, 1828**

***Arachotia* Moore, 1879**

Type species: *Arachotia flaviplaga* Moore; *Descr. Indian lep. Atkinson* (1): 14

88. *Arachotia euglenia* Jordan, 1908: 50

Type Locality: Khasia mountains, Assam (Jordan, 1908)

Distribution: India: Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

89. *Arachotia flaviplaga* Moore, 1879: 14

Type Locality: Darjiling (Moore, 1879a)

Distribution: India: Meghalaya, Nagaland, West Bengal (Joshi *et al*., 2021)

Host Plant: DD

90. *Arachotia vespoides* Moore, 1879: 390

Type Locality: North India (Moore, 1879b)

Distribution: India: Meghalaya (Joshi *et al.*, 2021)

Host Plant: DD

***Artona* Walker, 1854**

Type species: *Artona discivitta* Walker; *List Spec. Lepid. Insects Colln Br. Mus*. 2: 439

91. *Artona digitata* Hampson, 1919: 273

Type Locality: North India (Hampson, 1919)

Distribution: India; Myanmar (Burma) (Tenasserim) (Efetov and Tarmann, 2024)

Host Plant: DD

92. *Artona discivitta* Walker, 1854: 440

Type Locality: Nilgiris (Western Ghat Mountains, Tamil Nadu, southern India) (Walker 1854)

Distribution: India: Southern India (Efetov and Tarmann, 2024)

Host Plant: Poaceae (bamboo) (Efetov and Tarmann, 2024)

93. *Artona flaviciliata* Hampson, 1919: 273

Artona flaviciliata Hampson, 1919; *Novit. zool*. 26 (2): 273

Type Locality: Sikhim, Raitdong (Hampson, 1919)

Distribution: India: Sikkim (Efetov and Tarmann, 2024)

Host Plant: DD

94. *Artona flavigula* (Hampson, 1896): 477 (*Chrysartona*)

Type Locality: Khasis (Hampson, 1896)

Distribution: India: Assam (Efetov and Tarmann, 2024)

Host Plant: DD

95. *Artona flavipuncta* Hampson, 1900: 225

Type Locality: Khasis (Hampson, 1900)

Distribution: India: Assam (Efetov and Tarmann, 2024)

Host Plant: DD

96. *Artona fulvida* Butler, 1876: 356

= *Artona fulvida* var. *diffusa* Oberthür, 1894: 30

Type Locality: Moulmein (Butler, 1876)

Distribution: India: Northern; Myanmar (Burma), Thailand, Malaysia (Efetov and Tarmann, 2024)

Host Plant: DD

97. *Artona hypomelas* Jordan, 1907: 43

Type Locality: Mandi, NW.India and Chumbi Valley, Sikkim (Jordan, 1907)

Distribution: India: Sikkim (Efetov and Tarmann, 2024)

Host Plant: DD

98. *Artona microstigma* Jordan, 1907: 44

Type Locality: Khasia, Assam (Jordan, 1907)

Distribution: India: Assam (Efetov and Tarmann, 2024)

Host Plant: DD

99. *Artona phaeoxantha* Hampson, 1919: 274

Type Locality: Madras, Shevaroy Hills (Hampson 1919)

Distribution: India: Tamil Nadu (Efetov and Tarmann, 2024)

Host Plant: DD

100. *Artona posthyalina* (Hampson, [1893]): 244

Type Locality: Sikhim (Hampson, [1893])

Distribution: India: Sikkim (Hampson, [1893])

Host Plant: DD

101. *Artona sikkimensis* Elwes, 1890: 379

Type Locality: Singalelah (Elwes, 1890)

Distribution: India: Sikkim (Singh and Sheikh, 2024)

Host Plant: DD

102. *Artona (Zeuxippa) walkeri* (Moore, 1859): 199 (*Syntomis*)

≡ *Artona walkeri* (Moore, 1859): 43

Type Locality: Java (Moore, 1859)

Distribution: India: Himachal Pradesh, Arunachal Pradesh, Nagaland, Punjab, Sikkim; China, Indonesia, Malaysia, Myanmar (Pathania *et al*., 2021)

Host Plant: DD

103. *Artona (Zeuxippa) zebraica* Butler, 1876*:* 356

Type Locality: Almorah, massuri, N. India (Butler, 1876)

Distribution: India: Himachal Pradesh, Sikkim, Uttarakhand (Pathania *et al*., 2021)

Host Plant: *Oryza sativa* (Poaceae) (Robinson *et al*., 2010)

***Thibetana* Efetov & Tarmann, 1995**

Type-species: *Artona sieversi* Alphéraky, 1892: 5.

104. *Thibetana postalba (*Elwes, 1890): 379 (*Artona*)

Type Locality: Singalelah (Elwes, 1890)

Distribution: India: Sikkim (He *et al*., 2024)

Host Plant: DD

105. *Thibetana zebra* (Elwes, 1890): 379 (*Artona*)

Type Locality: Singalelah (Elwes, 1890)

Distribution: India: Sikkim (Singh and Sheikh, 2024)

Host Plant: DD

***Amuria* Staudinger, 1887**

Type species: *Amuria cyclops* Staudinger; *in Romanoff, Mém. Lépid*. 3: 172

106. *Amuria chorista* (Jordan, 1907): 44 (*Artona*)

Type Locality: Khasia, Assam (Jordan, 1907)

Distribution: India: Northeastern (Meghalaya) (Joshi *et al.*, 2021; Efetov and Tarmann, 2024)

Host Plant: Zingiberaceae (Efetov and Tarmann, 2024)

107. *Amuria lugubris* (Jordan, 1907): 44 (*Artona*)

Type Locality: Khasia Hills, Assam (Jordan, 1907)

Distribution: India: Northeastern (Meghalaya) (Joshi *et al.*, 2021; Efetov and Tarmann, 2024)

Host Plant: DD

108. *Amuria postvitta* (Moore, 1879): 13 (*Artona*)

Type Locality: Darjiling (Moore, 1879a)

Distribution: India: Sikkim (Efetov and Tarmann, 2024)

Host Plant: DD

109. *Amuria quadrimaculata* (Moore, 1879): 390 (*Brachartona*)

≡ *Artona quadrimaculata* (Moore, 1879): 390

Type Locality: Masuri, N.W. Himalayas (Moore, 1879b)

Distribution: India: Himachal Pradesh, Uttarakhand, West Bengal; Myanmar (Burma) (Pathania *et al*. 2021; Efetov and Tarmann, 2024)

Host Plant: DD

Chrysartona **Swinhoe, 1892**

Type species: *Procris stipata* Walker; *Cat. Het. Mus. Oxford* (1): 57

110. *Chrysartona* (*Chrystremewana*) *birmana* Efetov, 2006: 37

Type Locality: Burma, Bernardmyo (Efetov, 2006)

Distribution: India: Assam, Myanmar (Burma), Laos (Efetov and Tarmann, 2024)

Host Plant: DD

111. *Chrysartona* (*Chrysartona*) *efetovi* Parshkova, 2007: 143

Type Locality: North-eastern India

Distribution: India: Northeastern (Assam) (Efetov and Tarmann, 2024)

Host Plant: DD

112. *Chrysartona* (*Chrystremewana*) *honeyi* Efetov, 2006: 39

Type Locality: India, Khasis (Efetov, 2006)

Distribution: India: Northeastern (Assam) (Efetov and Tarmann, 2024)

Host Plant: DD

113. *Chrysartona* (*Chyrstarmanna*) *margarita* Efetov, 2006: 47

Type Locality: India, Margarita, Uppaer Assam (Efetov, 2006)

Distribution: India: Northeastern (Assam) (Efetov and Tarmann, 2024)

Host Plant: DD

114. *Chrysartona* (*Chrystarmanna*) *sikkima* Efetov, 2006 : 42

Type Locality: India, Sikkim (Efetov, 2006)

Distribution: India: Northeastern (Sikkim) (Efetov and Tarmann, 2024)

Host Plant: DD

115. *Chrysartona refulgens* Hampson, [1893]: 232

= *Artona refulgens* (Hampson, 1892): 43

Type Locality: Matelei, Burma (Hampson [1893])

Distribution: India; Myanmar, Thailand (Hampson, [1893])

Host Plant: DD

116. *Chrysartona stipata* (Walker, 1854): 114 (*Procris*)

≡ *Clelea stipata* (Walker, 1854): 46

Type Locality: North India

Distribution: India: Northern and Northeastern India; Myanmar (Burma) (Efetov and Tarmann, 2024)

Host Plant: *Ausonis japonica* (Vitaceae) (Robinson *et al*., 2010)

117. *Chrysartona* (*Chrysartona*) *tremewani* Efetov, 2006 : 31

Type Locality: India, Khasia Hills, Assam (Efetov, 2006)

Distribution: India: Northeastern India (Efetov and Tarmann, 2024)

Host Plant: DD

***Clelea* Walker, 1854**

Type species: *Clelea sapphirina* Walker; *List Spec. Lepid. Insects Colln Br. Mus*. 2: 465

118. *Clelea discriminis* Swinhoe, 1891: 475

Type Locality: Khasis (Swinhoe, 1891)

Distribution: India: Northeastern (Assam, Meghalaya, Nagaland) (Joshi *et al*., 2021; Efetov and Tarmann, 2024)

Host Plant: DD

119. *Clelea metacyanea* Hampson, 1896: 46

Type Locality: Khasia Hills (Hampson, 1896)

Distribution: India: Northeastern (Assam, Meghalaya) (Joshi *et al.*, 2021; Efetov and Tarmann, 2024)

Host Plant: DD

120. *Clelea nigroviridis* Elwes, 1890: 380

Type Locality: Naga Hills (Elwes, 1890)

Distribution: India: Northeastern (Nagaland); Myanmar (Burma) (Joshi *et al.*, 2021; Efetov and Tarmann, 2024)

Host Plant: DD

121. *Clelea plumbeola* Hampson, [1893]: 24

Type Locality: Bernardmyo, Burma (Hampson, [1893])

Distribution: India: Northeast (Assam, Meghalaya); Bhutan, Myanmar (Burma) **(**Joshi *et al.*, 2021; Efetov and Tarmann, 2024)

Host Plant: DD

122. *Clelea refulgens* Hampson, 1905: 193

Type Locality: Manipur, Mao (Hampson, 1905)

Distribution: India: Northeast (Assam, Manipur, Meghalaya); Myanmar (Burma) **(**Joshi *et al.*, 2021; Efetov and Tarmann, 2024)

Host Plant: DD

123. *Clelea sapphirina* Walker, 1854: 465

Type Locality: Hong Kong (Walker, 1854)

Distribution: India: Meghalaya; Southern China, Myanmar (Burma) (Joshi *et al.*, 2021; Efetov and Tarmann, 2024)

Host Plant: DD

124. *Clelea simplex* Jordan, 1908: 45

Type Locality: Khasia, Assam (Jordan, 1908)

Distribution: India: Northeastern (Assam, Meghalaya) (Joshi *et al.*, 2021; Efetov and Tarmann 2024)

Host Plant: DD

***Ephemeroidea* Hampson, [1893]**

Type species: *Ephemeroidea ariel* Hampson; *Fauna Br. India (Moths)* 1: 242

125. *Ephemeroidea ariel* Hampson, [1893]: 242

Type Locality: Bernardmyo, Burma (Hampson, [1893])

Distribution: India: Northeastern (Assam); Myanmar (Burma) (Efetov and Tarmann, 2024)

Host Plant: Poaceae (bamboo) (Efetov and Tarmann, 2024)

126. *Ephemeroidea cyanea* Jordan, 1908: 50

Type Locality: Khasia, Assam (Jordan, 1908)

Distribution: India: Northeastern (Assam, Meghalaya) (Joshi *et al.*, 2021; Efetov and Tarmann, 2024)

Host Plant: Poaceae (bamboo) (Efetov and Tarmann, 2024)

***Lophosoma* Swinhoe, 1892**

Type species: *Syntomis cuprea* Walker; *Cat. Het. Mus. Oxford* (1): 58

127. *Lophosoma cuprea* (Walker, 1856): 1596 (*Syntomis*)

Type Locality: Hindostan (India) (Walker, 1856)

Distribution: India: Andaman Islands, Himachal Pradesh, Sikkim, Meghalaya (Pathania *et al*., 2021; Joshi *et al*., 2021)

Host Plant: DD

128. *Lophosoma quadricolor* (Walker, 1856): 1596 (*Syntomis*)

Type Locality: Hindostan (India) (Walker, 1856)

Distribution: India: Northeastern (Efetov and Tarmann, 2024)

Host Plant: DD

***Hedina* Alberti, 1954**

Type species: *Northia tenuis* Butler; *Mitt. zool. Mus. Berl*.: 249

129. *Hedina tenuis* (Butler, 1877): 394 (*Northia*)

≡ *Illiberis tenuis* (Butler, 1877): 253

= *Northia khasiana* Moore, 1879: 12

≡ *Phacusa khasiana* (Moore, 1879): 49

Type Locality: Khasia Hills (Moore, 1879a)

Distribution: India: Meghalaya, Nagaland (Joshi *et al*., 2021)

Host Plant: Vitaceae, *Vitis amurensis*, *Vitis vinifera*

***Phacusa* Walker, 1854**

Type species: *Glaucopis tenebrosa* Walker; *List Spec. Lepid. Insects Colln Br. Mus*. 1: 150

130. *Phacusa dolosa* (Walker, 1856): 1594 (*Syntomis*)

Type Locality: North India (Walker, 1856)

Distribution: India: Assam (Joshi *et al*., 2021)

Host Plant: DD

131. *Phacusa properta* (Swinhoe, [1890]): 400 (*Notioptera*)

= *Northia dohertyi* Oberthür, 1894: 30

Type Locality: Rangoon Burma (Swinhoe, [1890])

Distribution: India; Myanmar (Burma) (Efetov and Tarmann, 2024)

Host Plant: DD

132. *Phacusa tenebrosa* (Walker, 1854): 150 (*Glaucopis*)

= *Phacusa siamensis* Oberthür, 1894: 31

≡ *Phacusa nicobarica* Hampson, 1920: 272

Type Locality: North India (Walker, 1854)

Distribution: India: Northeastern; Thailand (Efetov and Tarmann, 2024)

Host Plant: Vitacae, Dilleniaceae (Dillenia) (Küppers and Buchsbaum, 2015)

***Piarosoma* Hampson, [1893]**

Type species: *Piarosoma albicinctum* Hampson; *Fauna Br. India* (Moths) 1: 243

133. *Piarosoma arunachalensis* Sondhi, Efetov, Tarmann and Kunte, 2023: 139

Type Locality: Tale Wildlife Sanctuary, Lower Subansiri District, Arunachal Pradesh, India (Sondhi *et al*., 2023)

Distribution: India: Arunachal Pradesh (Sondhi *et al*., 2023)

Host Plant: DD

***Phacusamima* Efetov & Tarmann, 2025**

Type species: *Phacusa sizala* Swinhoe, 1894: 441

134. *Phacusamima sizala* (Swinhoe, 1894): 441(*Phacusa*)

≡ *Piarosoma sizala* (Swinhoe, 1894): 429–443

Type Locality: Shillong (Swinhoe, 1894)

Distribution: India: Meghalaya (Joshi *et al*., 2021)

Host Plant: DD

***Platyzygaena* Swinhoe, 1892**

Type species: *Soritia moelleri* Elwes; *Cat. Het. Mus. Oxford* (1): 57

135. *Platyzygaena moelleri* (Elwes, 1890): 385 (*Soritia*)

Type Locality: Sikkim? (Elwes, 1890)

Distribution: India: Assam (Joshi *et al*., 2021)

Host Plant: DD

***Tasema* Walker, 1856**

Type species: *Tasema bipars* Walker; *List Spec. Lepid. Insects Colln Br. Mus*. 7: 1597

136. *Tasema bipars* Walker, 1856: 1597

Type Locality: Hindostan (India) (Walker, 1856)

Distribution: India: Himachal Pradesh, Meghalaya, Nagaland (Pathania *et al.*, 2021)

Host Plant: DD

***Inope* Staudinger, 1887**

Type species: *Inope heterogyna* Staudinger; *in Romanoff, Mém. Lépid*. 3: 170

137. *Inope fuliginosa* (Moore, 1879): 14 (*Artona*)

Type Locality: Darjiling (Moore,1879a)

Distribution: India: West Bengal (Shah *et al*., 2021)

Host Plant: DD

***Thyrassia* Butler, 1876**

Type species: *Syntomis subcordata* Walker; *J. Linn. Soc. Lond. Zool*. 12 (60-62): 355;

138. *Thyrassia subcordata* (Walker, 1854): 132 (*Syntomis*)

Type Locality: North India (Walker, 1854)

Distribution: India: West Bengal (Shah *et al*., 2021)

Host Plant: DD

139. *Thyrassia virescens* (Hampson, [1893]): 238 (*Monoschalis*)

Type Locality: Colombo, Ceylon (Hampson, [1893])

Distribution: India: Karnataka (Mahendra and Samraj, 2022) Global: Sri Lanka (Hampson, [1893])

Host Plant: DD

Subfamily **Zygaeninae Latreille, 1809**

Praezygaena **Alberti, 1954**

Type species: *Zygaena myodes* Druce; *Mitt. zool. Mus. Berl*. 30 (2): 185

140. *Praezygaena caschmirensis* ([Kollar, 1844]) : 459 (*Zygaena*)

= *Zygaena asoka* Moore, 1879: 389; **Type Locality**: Kaschmir

Type Locality: Kashmir and Himalayas ([Kollar, 1844])

Distribution: India: Himachal Pradesh, Jammu Kashmir, Punjab; Pakistan, Nepal (Pathania *et al*. 2021; Sondhi *et al*., 2026)

Host Plant: Cuscuta (Convolvulaceae) (Robinson *et al*., 2010)

141. *Praezygaena erythrosoma* (Hampson, [1893]): 231 (*Zygaena*)

Type Locality: Almora (Hampson, [1893])

Distribution: India: Uttarakhand (Hampson, [1893])

Host Plant: DD

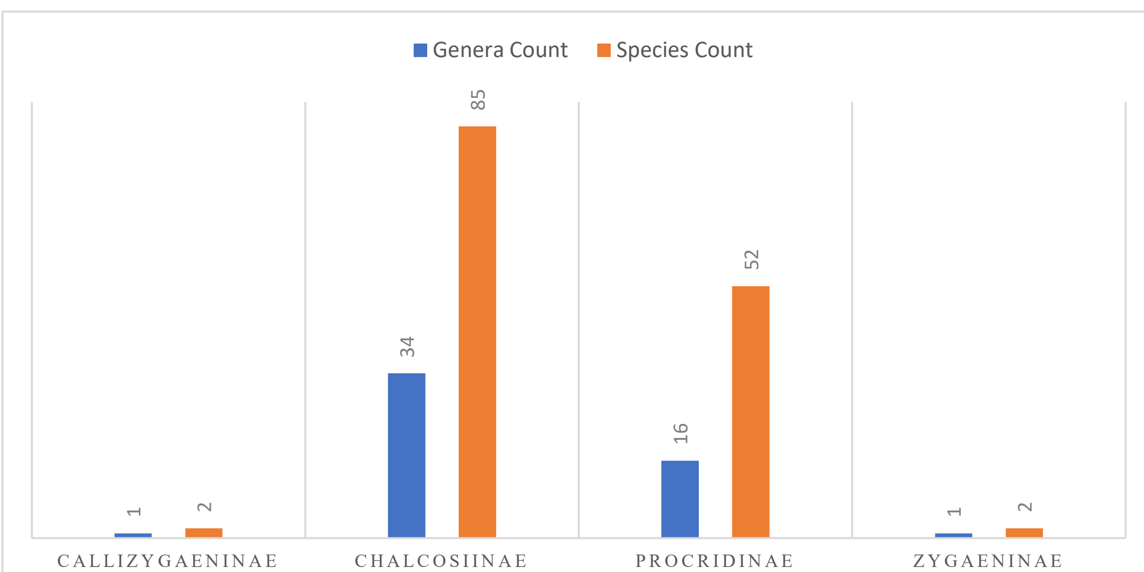

Fig. 1. Subfamily-wise species and genera count of Zygaenidae from India.

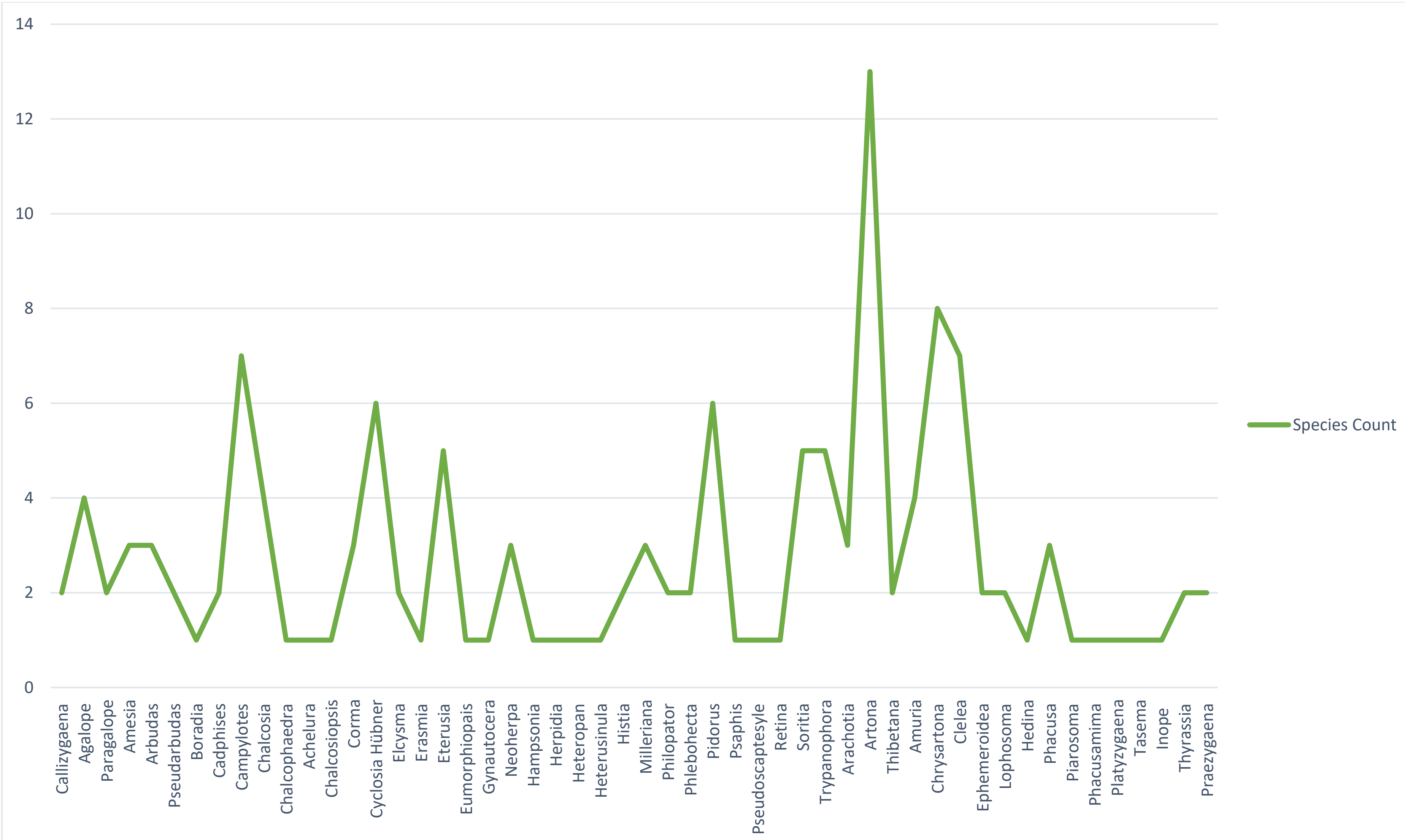

Fig. 2. Genera-wise species count of Zygaenidae from India.

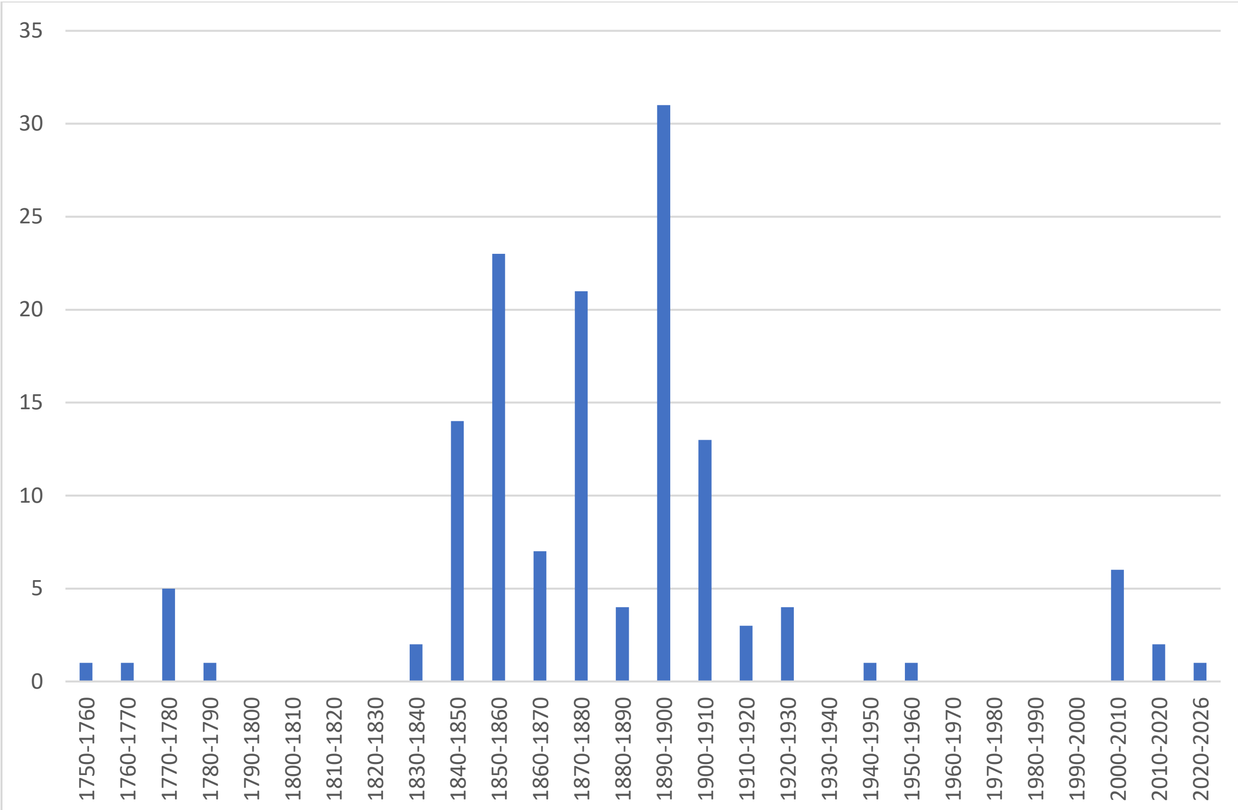

Fig. 3. Decade-wise species count of Zygaenidae from India.

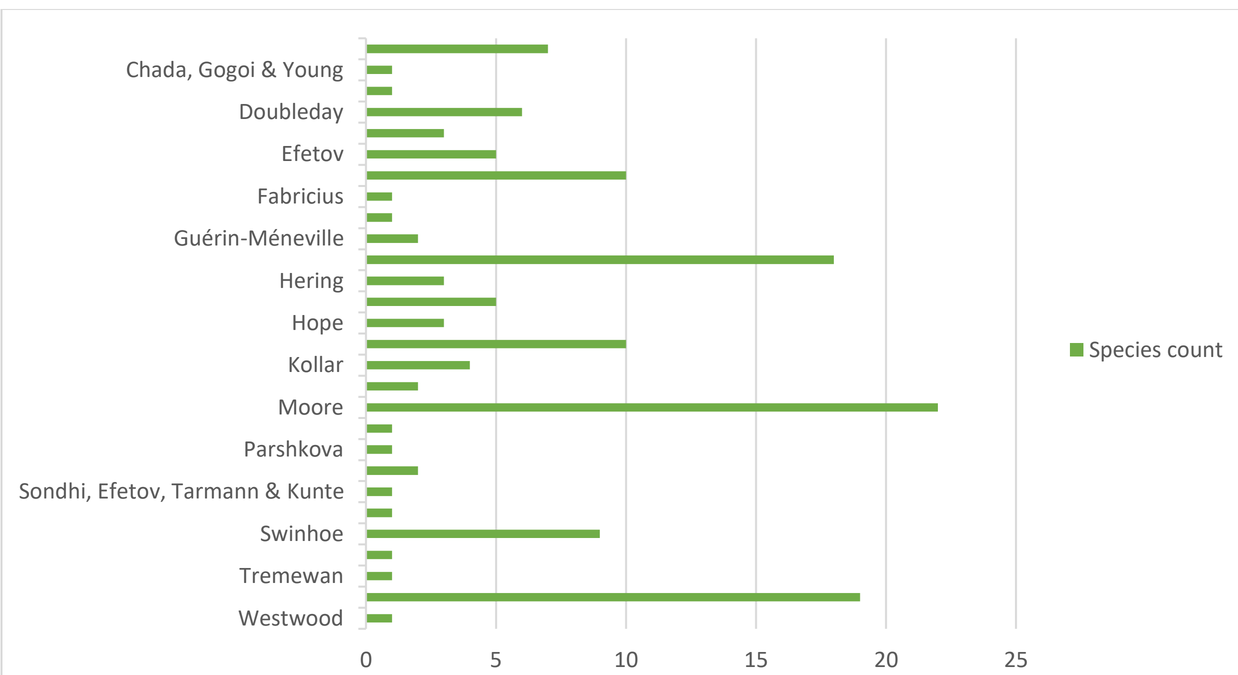

Fig. 4. Author and species count of Zygaenidae from India.

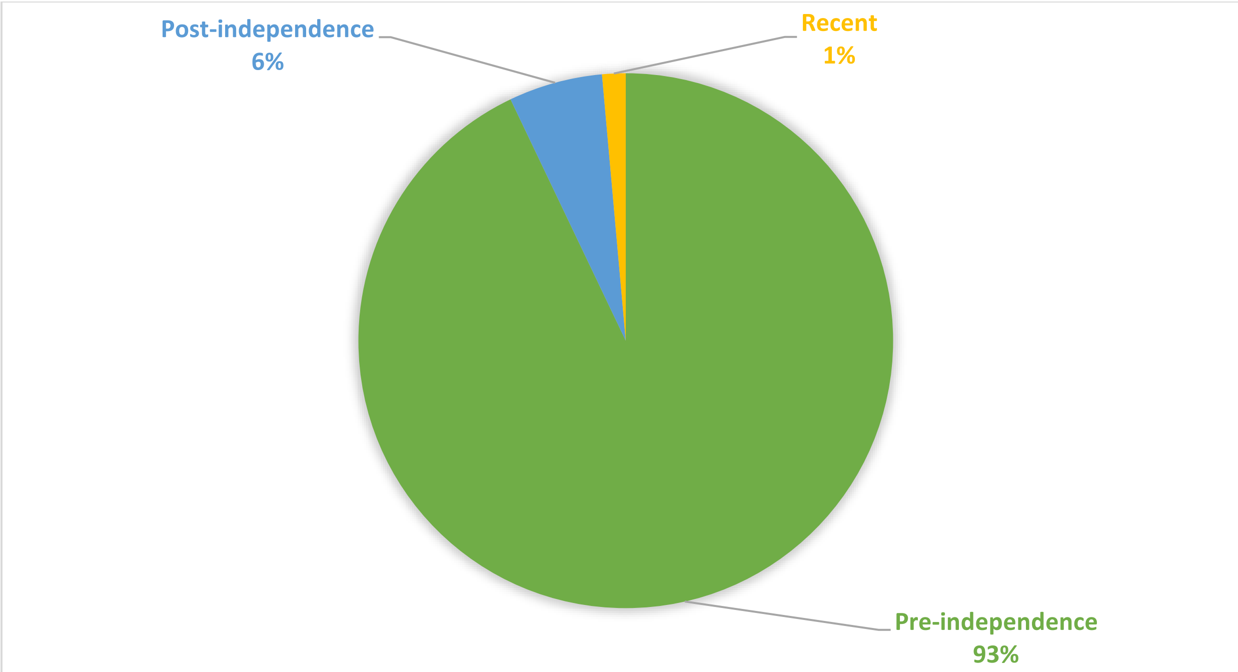

Fig. 5. Pre-independence, post-independence and recent data on the description of species from Zygaenidae.

Acknowledgements

The authors are thankful to the Director, Zoological Survey of India, Kolkata and the Officer-in-Charge, WRC, ZSI, Pune, for facilities and encouragement. The authors are thankful to the reviewer's and the subject editor for their valuable suggestions and constructive criticism that improved the manuscript.

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MS Received on 26th December, 2025
MS Accepted on 27th January, 2026

DOI: 10.55278/POMF7096

Antheraea cingalesa (Moore, 1883) (Lepidoptera: Saturniidae): First Verified Laboratory Rearing and Life-History Documentation in Sri Lanka

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Abstract

The Family Saturniidae comprises medium to very large sized moth species, characterized by robust bodies coated with dense, hair like scales and in most species, prominent eyespots on the wings. Saturniidae includes species in 162 genera and nine subfamilies and is notable among Lepidoptera for its economic value as silk producers, agricultural pests and for their ornamental appearance. The Tusser silk moth is among the species from which wild silk is extracted. In India wide range of silk is produced using close relatives of *Antheraea cingalesa*, which is endemic to Sri Lanka and first described by Moore (1883). However there are no records of silk production using this species in Sri Lanka.

This study reports the successful laboratory rearing of field-collected *A. cingalesa* in Jaffna, located in the Northern Province of Sri Lanka, from October to December 2024, culminating in the emergence of viable adults. Although Jaffna lies within the dry zone and receives monsoonal rainfall from October to January, periodic temperature fluctuations can adversely affect developmental stages, highlighting the importance of stable environmental conditions for effective rearing. The results of this study indicate that *A. cingalesa* holds promising potential for future commercial or small-scale silk production in the region.

Keywords: Saturniidae, *Antheraea cingalesa*, Tusser silk moth, laboratory rearing

Introduction

The family Saturniidae is a diverse group of moths within the order *Lepidoptera*, comprising medium- to very large-sized species. These moths are typically crepuscular and are characterized by robust bodies covered with dense, hair-like scales. Most species possess prominent wing eyespots, which serve as anti-predatory adaptations. Members of this family lack a functional proboscis, and their labial palpi are extremely small. Both sexes have bipectinate antennae, with males exhibiting notably longer branches that reach maximum length at mid-antenna before tapering toward the base and tip. Their legs are short, hairy, and devoid of spurs (Butler, 1892).

According to the Classification proposed by Lemaire and Minet (1998), Family Saturnidae is containing 1861 species in 162 genera and nine subfamilies (Regier *et al*., 2008). Most notable species including the emperor moths, royal moths and giant moths belong to this family. They are notable among Lepidoptera for their economic value as silk producers, agricultural pests and for their ornamental appeal (Aluthwattha, 2013). The cocoons of saturniids are used for the production of wild silk, and in some larvae or pupae of selected species play a role in human nutrition. Their large body size has made saturniids particularly useful as model organisms in comparative studies of genetics, developmental biology, and physiology, and ecology and behavior (Regier *et al*., 2008).

The earliest documentation of Sri Lankan Saturniidae dates back to Tennent (1861), who recorded three species: the Moon moth, Atlas moth, and Tussar moth. Moore (1882) later redescribed these species under their scientific names—*Actias selene* Hübner, *Attacus taprobanis* Moore, and *Antheraea cingalesa* Moore. *Antheraea cingalesa*, a tussar silk moth endemic to Sri Lanka, was formally described by Moore (1883). Despite this, it has been frequently misidentified in later literature as *Antheraea mylitta* or *Antheraea paphia*. Current phylogenetic evidence, however, supports *A. cingalesa* as a distinct and valid endemic species. Although India produces a wide range of wild silks using species closely related to *A. cingalesa*, no reliable records exist of its use in silk production within Sri Lanka.

Sri Lanka's silk industry dates back to the Dutch–Ceylon period, centered around the mulberry silk moth *Bombyx mori* (Family: Bombycidae). Established as a government-sponsored plantation sector, the industry experienced periods of expansion and decline. Between 1980 and 1992, over 400 acres were developed for sericulture, and 16 government-run farms operated successfully in regions such as Pallekala, Vaddakkachchi, Iranamadu, Korana, Galaha, and Battigama (Pakeerathan, 2023). Despite the industry's promising performance, mulberry silk production was discontinued in 1998 due to socio-religious concerns and the increasing availability of inexpensive synthetic silk (Pakeerathan, 2023; Wijesiri, 2020). Currently, Sri Lanka imports raw silk and silk yarn for its textile sector, even though many private enterprises continue to manufacture silk-based products. Reviving domestic silk production could reduce costs, strengthen international competitiveness, and contribute positively to the national economy (Wijesiri, 2020). In this context, assessing the potential of the naturally occurring *Antheraea cingalesa* offers a promising avenue for re-establishing a sustainable wild-silk industry in Sri Lanka.

Accordingly, the present study was conducted in 2024 at the Department of Zoology, University of Jaffna, to document the life history of *Antheraea cingalesa* and evaluate its feasibility for commercial or small-scale indoor rearing as a foundation for future economic development.

Methodology

During a field survey on 26 October 2024, a pair of mature *Antheraea cingalesa* adults (one male and one female) were collected from a *Syzygium cumini* tree (Tamil: *Naaval* (நாவல் மரம்); Sinhala: *Madan* (මාදං)) near Uppuvayal Kulam, Arali, Jaffna (9°43'32.3"N, 79°57'03.0"E) (Fig. 1-a). The adult pair was transported to the laboratory of the Department of Zoology, University of Jaffna, and maintained under indoor conditions for mating and oviposition.

The eggs laid by the female were collected and placed in separate plastic containers (Fig 1-b) lined with tender leaves of *S. cumini* to allow incubation under ambient laboratory conditions until hatching. Upon emergence, the larvae were divided into groups and reared in insect rearing cages. Fresh leaves of *Syzygium cumini* and *Terminalia arjuna*—a reported host plant (Aluthwattha, 2013)—were provided throughout the larval development until cocoon formation.

After pupation, the cocoons were collected and suspended inside the rearing cage (Fig 4-a) along with *T. arjuna* foliage to maintain a natural microenvironment. All rearing procedures were conducted under indoor laboratory conditions, with temperature maintained at 24–29°C, relative humidity (RH) between 70–80%, and a 12L: 12D photoperiod.

Emerging adults were collected, identified to species level, and their morphological characteristics were recorded. Specimens were then properly pinned, dried, and preserved in insect storage boxes for further examination.

Results and Discussion

Antheraea cingalesa was first documented in the Jaffna District of Sri Lanka's Northern Province from the Velanai area in 2015. Subsequent isolated findings included a single cocoon collected from *Syzygium cumini* in Kokuvil in 2018 and another from *Pemphis acidula* in the Mandaitivu mangrove ecosystem in 2023. In both cases, the cocoons emerged into single female adults, which deposited unfertilized eggs, preventing the establishment of a colony.

In the present study, a field-collected, already-mated female deposited 120 eggs within two days. Of these, 80 eggs hatched, yielding a 67% hatching rate. The first-instar larvae showed a clear feeding preference for

Terminalia arjuna leaves, while *S. cumini* leaves were largely rejected. Among the 80 larvae, only 12 individuals (10%) survived to maturity and spun cocoons. The first cocoon was initiated 33 days after larval emergence, and the remaining larvae completed cocoon formation within 45 days. Adult emergence occurred 57–70 days after pupation.

Of the 12 cocoons, only 5 adults (4%) successfully emerged—three males and two females—indicating an overall survival rate of 4% from egg to adult. Completing the entire first generation required approximately 108 days.

Despite the high initial egg production, larval mortality was recorded at all developmental stages. This appears to be largely associated with suboptimal indoor laboratory conditions. Under natural conditions, tussar moth larvae occupy the sheltered spaces between fresh leaves, which provide a stable and humid microclimate (Liu et al., 2025). In the laboratory, although fresh leaves were provided regularly, they dried within hours, making it difficult to mimic natural microhabitat stability. Frequent replacement helped but did not fully recreate the buffering provided by living foliage. Additionally, even minor fluctuations in temperature and humidity under laboratory conditions may have contributed to the high mortality observed.

Adult emergence followed the typical pattern observed in saturniid moths: males emerged earlier, were active immediately, and initiated mating soon after the females emerged. Female moths began laying viable eggs within two days of mating, confirming the successful completion of the reproductive cycle under laboratory conditions.

Table 1: Life span of *Antheraea cingalesa* when reared in the laboratory (indoor) condition (October 2024-February 2025).

Life stages	Duration of different stages (Days) Mean ± SD
Egg	8 ± 0.82
Larva: 1st instar	6.5 ± 0.5
2nd instar	6 ± 0.82
3rd instar	5.53 ± 3.44
4th instar	6.5 ± 1.29
5th instar	6.0 ± 1.0
Cocoon	64 ± 8.35
Adult	6.5 ± 2.65

Although survival rates were low, this study represents the first successful laboratory rearing of *A. cingalesa* from field-collected adults to the emergence of a viable next generation in the Jaffna region. The high mortality highlights the need for improved environmental control—particularly in maintaining leaf freshness, humidity, and temperature consistency. With optimized rearing conditions, *A. cingalesa* shows promise as a potential candidate for future wild-silk production initiatives in Sri Lanka.

Description of life stages

Eggs

The eggs are pale yellow, ovoid, and dorsoventrally flattened. They measure 3.13 ± 0.17 mm in length and 2.87 ± 0.06 mm in breadth. A distinct dark brown lateral ring encircles each egg ((Fig. 2-b, c)).

Larva:

The larvae undergo four moults, passing through five instar stages. Newly hatched first instar larvae are brown, turning green by the second instar. The body bears multiple tubercles, each supporting clusters of setae (Fig. 3-a–e).

The mature larva exhibits:

- Two dorsal rows of orange-coloured tubercles, each tipped with a few short radiating hairs and accompanied by purple bases.
- A lateral row of eight eyespots presents on both sides, from the 1st to the 8th abdominal segments.
- Purple tubercles situated above and below each eyespot, extending from the 1st thoracic segment to the 8th or 9th abdominal segment.
- A yellow lateral stripe, broadening into a brown band on the anal segment.
- A small, rounded head capsule with three pairs of thoracic legs and five pairs of abdominal prolegs (on the 3rd–6th and 10th abdominal segments).

The larvae feed voraciously on *Terminalia arjuna* leaves throughout their development.

Cocoon

At the end of the final larval instar, the caterpillar constructs a protective silk cocoon. The cocoon is oval, firm, and brownish-grey, and is attached to a twig by a short, coarse silken stalk. The average cocoon length is 2.79 ± 0.97 cm. The cocoons are typically enclosed within leaves, which provide an additional layer of protection in natural settings (Fig: 4-b).

Pupa: (Fig: 4- d, e)

The pupa is ovoid, with a blunt anterior and a triangular posterior end, and is dark brown in colour. Segmentation is clearly visible, distinguishing the head, thorax, and abdomen.

Sexual dimorphism is evident at the pupal stage:

- Female pupae exhibit fine, longitudinal lines on the 8th and 9th abdominal segments.

- Male pupae have a distinct ventral dot on the 9th abdominal segment.

These features align with diagnostic characters described for *Antheraea mylitta* by Bambhaniya *et al*. (2017), supporting their taxonomic relevance in *A. cingalesa* as well.

Adults

Male: The male has a wingspan of 128 mm. The overall coloration is a deep reddish-ochre, with the forewings becoming a paler ochre near the apex, sprinkled with purplish-white scales. The costal margin appears hoary. A faint darker-ochre band crosses the disc through the ocellus, accompanied by a similar outer lunular shade (Fig. 5-b).

A slender purple-brown streak, lightly edged with grey, runs across the basal region of the cell, with another wavy sub-basal streak of the same hue positioned below it. A sub marginal purple-brown line is present, bordered externally by a faint purplish-white lunular band. The outer margin of the forewing shows a slight olive tint.

On the hind wing, a faint wavy purple-brown sub-basal transverse streak is visible. A very faint ochreous band crosses the disc through the cell, followed by a lunular outer discal shade. Beyond this lies a sub marginal lunular purple-brown line, edged externally with a barely perceptible purplish-white border.

The ocelli on both wings are round, consisting of a black outer ring, a purplish inner border, and a central white line.

Females:

The female is larger, with a wingspan of 144.6 mm. The body and wings are a rich yellowish-ochre, and the ocelli are larger and more prominently defined than in the male. The sub-basal streaks are clearer, and the sub marginal purple line is stronger and more distinct, broadly edged on the outer side with purplish-white.

The anterior thorax and collar are hoary, contrasting with the ochreous coloration of the head, palpi, and legs (Fig: 5-a).

Conclusion

The present study demonstrates that *Antheraea cingalesa* can be successfully reared under indoor, small-scale laboratory conditions in Jaffna when suitable environmental parameters are maintained. A temperature range of 24–29 °C and relative humidity of 70–80% were found to be conducive for improving larval survival and supporting successful completion of the life cycle. With appropriate technical guidance and the necessary infrastructure, the expansion of *A. cingalesa* culture holds promising potential for revitalizing Sri Lanka's silk industry. Such initiatives may reduce dependence on imported silk, lower production costs for silk-based products, and contribute to future economic development.

Table 2: Morphometric characteristics of life stages of *A. cingalesa* when reared in the laboratory (indoor) condition (October 2024-February 2025)

Stages		Measurements	Mean (mm) ± SD (N=5)
Egg		Length	3.13 ± 0.17
		Breadth	2.87 ± 0.06
Larva	1st instar	Length	13.93 ± 5.22
	2nd instar		27.93 ± 4.05
	3rd instar		43.43 ± 5.72
	4th instar		65.75 ± 4.35
	5th instar		80.5 ± 9.88
Cocoon	Male	Length	42.7 ± 0.58
		Breadth	23.7 ± 0.58
	Female	Length	48.8 ± 2.77
		Breadth	26.8 ± 2.28
Pupa	Male	Length	32.5 ± 0.71
		Breadth	16.5 ± 0.71
	Female	Length	40.0 ± 2.83
		Breadth	21.5 ± 0.71
Adult	Male	Length of the body	35.8 ± 2.77
		Breadth with wing expanded	128.0 ± 5.70
		Length of the antenna	13.6 ± 1.14
	Female	Length the body	38.4 ± 5.18
		Breadth with wing expanded	144.6 ± 13.90
		Length of the antenna	14.4 ± 1.14

9°43'31.7"N 79°57'05.2"E

Fig. 1: a. Collection locality of ***A. cingalesa*** at Arali, Jaffna. **b.** ***A. cingalesa*** adult Mating pair

Fig. 2: Oviposition and eggs of ***A.cingalesa***

a. Mating pair kept in the plastic box **b**: eggs in the plastic container **c-** eggs x 20.

Fig. 3: Larval stages of ***A. cingalesa***

a. Ist instar **b**: IInd instar **c-**. IIIrd instar **d**: IVth instar **e**. Vth instar larva

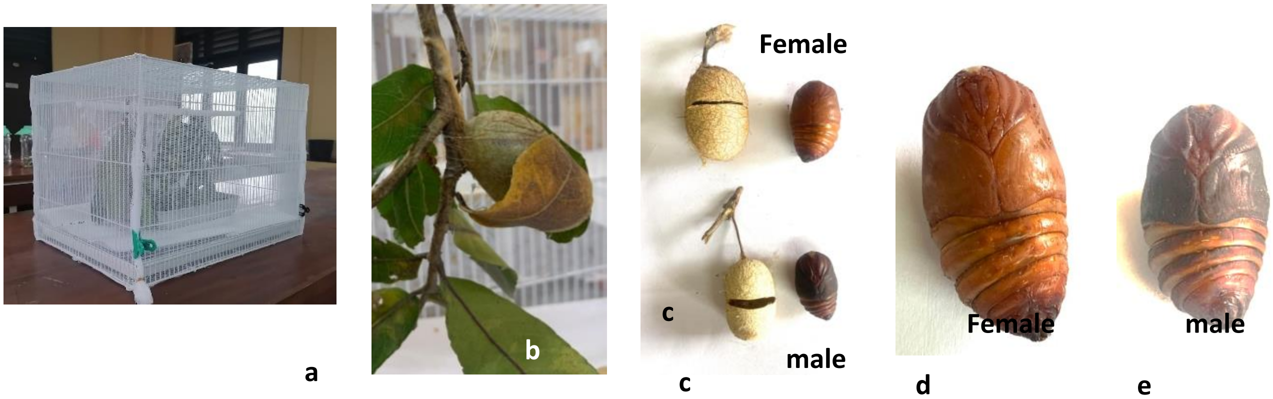

Fig. 4:

a Laboratory Rearing cage **b**: pupation **c**. cocoon and pupa **d**. female pupa **e.** male pupa

Fig. 5: Adult *Antherea cingalesa* – Habitus

a Female moth -Wing span 144.6 mm **b**: Male moth-wing span 128mm

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MS Received on 03rd January, 2026
MS Accepted on 5th February, 2026

DOI: 10.55278/WUNM4171

First Documentation of Two *Odontotermes* Species (Blattodea: Termitidae) from Assam, India

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Abstract

Termites represent one of the most ecologically and economically important groups of insects, yet their true diversity in northeastern India remains insufficiently explored. The genus *Odontotermes* is among the most species-rich in the Oriental region, but many of its members remain poorly documented due to their cryptic habits and overlapping morphological traits. Assam, despite its rich biodiversity and varied forest ecosystems, has historically lacked comprehensive faunistic surveys of this genus. In this study, two termite species—*Odontotermes annulicornis* Xia & Fan, 1982 and *Odontotermes foveafrons* Xia & Fan, 1982 (Blattodea: Termitidae)—are recorded for the first time from Assam, India. Both species were reported only recently from India and were otherwise known solely from their type localities in Yunnan, China. Diagnostic notes, locality information, and brief comparative remarks are provided to support accurate identification. These new state records expand the known Indian distribution of *Odontotermes* and contribute valuable baseline data for future faunistic, ecological, and applied research in northeastern India, a region where termite diversity remains under-documented but highly significant from both ecological and developmental perspectives.

Key words: *Odontotermes*, Assam, new records, morphometric measurements

Introduction

Termites are highly eusocial insect group with clear hierarchy and sharp division of labor, living in colonies with either single nest or several interconnected nests (Ningthoujam et al., 2024). They are the most dominant arthropod decomposers in the tropical forests (Wood and Sands, 1978; Matsumoto and Abe, 1979; Collins, 1983). They are abundant throughout the tropics and subtropics, as well as in many temperate areas of world, with species diversity and total biomass being greatest in the tropics (Pearce, 1997). Factors like vegetation, food availability and soil type influence the mound-building behavior and distribution of termites (Korb, 2010), at regional level.There are 3176 known species (2976 living and 200 fossils) (Constantino 2021) of which 325 species under 53 genera and 8 families are found in India (Baraik et al., 2025; Roy et al., 2025). Genus *Odontotermes* Holmgren, 1910, (Family Termitidae) comprises 45 species in India (Roy et al., 2025), amounting to almost 13.7% of the total termite species. Termitidae is the most species rich family among the extant family of termites (Hellemans et al., 2024). In this study we report the first time occurrence of two species of termites belonging to family Termitidae from Dhemaji and Lakhimpur districts of Assam.

Material and Methods

Samples were collected from Dhemaji and Lakhimpur districts of Assam. The classification system followed Hellmans et al. (2024), while Chhotani (1997) and Xia and Fan (1982) were consulted for generic and species-level identification.

All collected specimens were preserved in absolute alcohol immediately after collection. Morphometric measurements were taken under a Leica EZ4 stereo microscope with specimens submerged in alcohol to maintain structural integrity. High-resolution images were captured using a Leica M205A stereo microscope equipped with a Leica DFC500 camera, and processed using the extended-focus software LAS Version 3.6.

Preservation and Deposition

After examination, all specimens were transferred to fresh absolute alcohol and deposited in the National Zoological Collection, Zoological Survey of India (ZSI), Kolkata, for long-term preservation and reference.

Results:

***Odontotermes annulicornis* Xia and Fan, 1982**

Material examined: 1 vial containing 45 soldiers from Dhemaji, Assam (Lat. 27.4800N; Long 94.5800E) 19.ix.2025, coll. Kangkana Lekharu, ex. near human habitation associated with paddy fields.

Diagnostic Characters (Table 1): Head nearly oval with maximum width at the posterior end. Antennae with 16 segments; 4th segment smallest. Lateral margin of postmentum convex, with maximum width in the middle. Mandibles slender with incurved tip.

Table 1: Measurements (mm) of soldiers of *Odontotermes annulicornis* Xia and Fan, 1982

Characters	Xia and Fan, 1982	Present Study
Head length to lateral base of mandibles	1.25-1.50	1.40-1.45
Maximum head width	1.08-1.28	1.15-1.20
Head width at the base of mandibles	0.59-0.72	0.60-0.62
Mandible length	0.78-0.93	0.90
Tooth distance	0.22-0.28	0.23-0.25
Tooth index (Tooth distance / Mandible length)	0.26-0.30	0.26-0.27
Head width / Head length index	0.82-0.89	0.82-0.85
Head mandible index	0.62	0.62-0.64
Antennal Segments	16	16

Odontotermes annulicornis Xia and Fan, 1982

Distribution: India: Gujarat, Maharashtra (Roy et al., 2025) and Assam (new record). Type-locality: Jinghong, Yunnan, China (Krishna et al. 2013)

Remarks: The pest status of the species is unknown.

***Odontotermes foveafrons* Xia and Fan, 1982**

Material examined: A. 1 vial containing 5 soldiers and 25 workers from Dhemaji, Assam (Lat. 27.434713N; Long 94.347934E) 28.ix.2025, coll. Kangkana Lekharu, ex. near human habitation. **B.** 1 vial containing 24 soldiers and 15 workers from Panigaon, Lakhimpur district, Assam (Lat. 27.14995N; Long 94.112976 E) 05.x.2025, coll. Kangkana Lekharu, ex. near human habitation. **C.** 1 vial containing 4 soldiers and 20 workers from Ranganadi Lakhimpur, Assam (Lat. 27.127721N; Long 94.03105E) 03.x.2025, coll. Kangkana Lekharu, ex. near human habitation, **Diagnostic Characters (Table 2):** Head is subrectangularly oval with maximum width of head is in the middle. Mandibles are stouter with incurved tips. Antennae with 16 segments; segment 4 smallest.

Table 2: Measurements (mm) of soldiers of *Odontotermes foveafrons* Xia and Fan, 1982

Characters	Xia and Fan, 1982	Present Study
Head length to lateral base of mandibles	1.19-1.47	1.25-1.35
Maximum head width	1.03-1.23	1.10-1.15
Head width at the base of mandibles	0.61-0.77	0.70-0.75
Mandible length	0.72-0.88	0.75-0.80
Tooth distance	0.19-0.25	0.20
Tooth index (Tooth distance / Mandible length)	0.25-0.29	0.25-0.26
Head width / Head length index	0.81-0.88	0.85-0.88
Head mandible index	0.59-0.60	0.57-0.62
Antennal Segments	16	16

Odontotermes foveafrons Xia and Fan, 1982

Distribution: India: Maharashtra (Roy et al., 2025) and Assam (new record). Type-locality: Yangwu, Yunnan, China (Krishna et al. 2013).

Remarks: The pest status of the species is unknown. The species was found in association with Indian cork (*Millingtonia hortensis*), mango (*Mangifera indica*), and jackfruit (*Artocarpus heterophyllus*).

Discussion:

Genus *Odontotermes* Holmgren has distribution in Ethiopian and Oriental regions and is the most widely distributed and prolific species of termites in India (Chhotani, 1997). Out of 36 species of termites reported from Assam earlier, 12 species belonged to the genus *Odontotermes* Holmgren, 1910 (Buragohain et al., 2025). The species *O. annulicornis* and *O. foveafrons* were previously known only from their type-localities in China (Xia & Fan 1982). However, both the species were recorded recently in India, *O. annulicornis* from Gujarat and Maharashtra and *O. foveafrons* from Maharashtra (Roy et al., 2025) and is now reported from Assam through the present study.The species can be distinguished from all the previously known species of *Odontotermes* in India based on their morphometry (Table 1 and Table 2).They are closely related to *Odontotermes obesus* (Rambur, 1842), but differ in head shape, mandible morphology and tooth index (Roy et al., 2025). In India, under *Odontotermes*, 8 species are categorized as major pests, while 13 are minor pests (Rajmohana et al., 2019 and Shanbhag &Sundararaj, 2013), but the pest status of *O.annulicornis* and *O. foveafrons* remain unknown. In-depth, seasonally surveys combined with extensive taxonomic investigations are likely to reveal additional termite diversity, including previously unrecorded species and new distributional records in India.

Conclusion

The present study documents *Odontotermes annulicornis* and *O. foveafrons* from Assam for the first time, thereby extending their known distribution within India beyond Gujarat and Maharashtra. These findings not only update the termite checklist of the state but also highlight the presence of poorly known species previously restricted to their type localities in Yunnan, China. Both species are morphologically distinguishable from related congeners based on diagnostic characters and morphometric measurements, underscoring the importance of careful taxonomic assessment in regional faunal studies.

Given that the pest status of both species remains unknown, and considering the ecological and economic significance of *Odontotermes* in agricultural and forest ecosystems, further seasonal and habitat-specific surveys are essential. Comprehensive studies integrating morphology, molecular diagnostics, and ecology will likely reveal additional species, refine distribution ranges, and contribute to a deeper understanding of termite diversity in northeastern India.

Acknowledgement

Authors[1&1*] are grateful to the director, Zoological Survey of India, Kolkata for encouragement and research facilities. The authors[2&3] are thankful to Dept. of Zoology, Namrup College, Dibrugarh, Assam and the Department of Entomology, Rajiv Gandhi University, for academic support during the course of this study.

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MS Received on 01st January, 2026
MS Accepted on 15th February, 2026

DOI: 10.55278/FSMX7943

A Severe Outbreak of the Ailanthus Webworm, *Atteva fabriciella* Swederus (Lepidoptera: Attevidae), on *Ailanthus triphysa* (Simaroubaceae) in Kerala, India: Taxonomy, Distribution, Biology and Damage Symptoms

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Abstract

A sudden and severe outbreak of the ailanthus webworm, *Atteva fabriciella* Swederus (Lepidoptera: Attevidae), was recorded on *Ailanthus triphysa* (Simaroubaceae) at Parakkadavu, Kozhikode district, Kerala, India, between 25 January and 1 February, 2026. Larvae constructed extensive silken leaf webs, resulting in pronounced defoliation and foliar damage. Field surveys revealed abundant caterpillars, pupae, and adult moths. Several individuals who visited the site reported intense itching after contact with dense larval aggregations, suggesting possible dermal irritation linked to larval setae. After feeding, larvae migrated to accumulated dried foliage on the ground or pupated among residual silk masses or under the leaves. The affected host tree was a mature specimen (~60 years old) with a large branching structure. This paper provides anan integrated account of the outbreak, including updated taxonomic context of *A. fabriciella*, along with global and regional distribution records, known host associations, key aspects of its biology, characteristic damage symptoms, and parasitoids reported from the species. The observations offer valuable baseline data for future monitoring and management of *A. fabriciella* in forestry, agroforestry, and urban landscape systems.

Keywords: *Atteva fabriciella*; Attevidae; *Ailanthus triphysa*; outbreak; taxonomy; defoliation; dermal irritation; parasitoids; India

Introduction

The genus *Atteva* Walker, 1854 is the sole representative of the pantropical moth family Attevidae within the superfamily Yponomeutoidea, comprising over 50 described species with highest diversity in the Indo-Australian tropics (Heppner 2009; Sohn & Wu 2013). Attevidae is characterized by morphological specializations such as reduced hind leg spurs, distinctive chaetosema on the head, and communal larval webbing on host plants (Sohn & Wu 2013).

Atteva fabriciella Swederus, 1787 (originally described as *Phalaena (Tinea) fabriciella*) is a cosmopolitan moth whose foliage-feeding larvae are oligophagous on hosts in the family Simaroubaceae, particularly *Ailanthus* spp. including *Ailanthus triphysa* and *A. excelsa* (Varma, 1986). Extensive synonymy and taxonomic confusion historically surrounded *A. fabriciella*, with names such as *Atteva brucea* Moore and *Corinea niviguttella* Walker now treated as junior synonyms following recent revisions and neotype designations (Sohn & Wu 2013).

Atteva fabriciella (Swederus, 1787) is a widely distributed species in the Oriental region, with records extending across much of South and Southeast Asia**.** Published distribution records indicate its occurrence in India, Sri Lanka, Bangladesh, Thailand, Vietnam, Malaysia, Indonesia (Java, Sumatra, Sulawesi, Borneo), the Philippines, and southern China (Sohn & Wu, 2013; Robinson et al., 1994). Recent faunistic surveys have further expanded its known distribution within the Oriental region, including new records from Cambodia, highlighting the continuing documentation of the species across Southeast Asia (Na & Bae, 2024). Within India**,** *A. fabriciella* has been reported from several states including Gujarat, Odisha, Assam, Meghalaya, and parts of peninsular India, where it is commonly associated with *Ailanthus* plantations and forest habitats (Robinson et al., 1994; Varma, 1986). In southern India**,** particularly Kerala**,** the species has been documented as a significant defoliator of *Ailanthus triphysa* in nurseries and young plantations, occasionally causing localized outbreaks (Varma, 1986). Overall, the species' distribution is strongly correlated with the geographic range of its host plants, particularly *Ailanthus* spp., which largely determines its regional abundance and pest status in tropical Asia (Sohn & Wu, 2013).

Severe defoliation by *A. fabriciella* has been documented mostly in plantations, nurseries, and occasionally on mature trees, detailed reports of outbreaks involving intense population buildup remain relatively scarce in peer-reviewed literature. This study describes a severe outbreak on a mature *A. triphysa* tree in Kerala, India, and emphasizes the taxonomic context, biological characteristics, damage symptoms, associated parasitoids etc.

Materials and Methods

Study Area and Host

The outbreak was recorded and monitored at Parakkadavu, Kozhikode district, Kerala, India (11.729227°N, 75.638593°E) during 25 January to 1 February, 2026. The host plant, Ailanthus triphysa (Simaroubaceae), was a mature tree approximately 60 years old, with a well-developed canopy and extensive branching. According to local accounts, this tree was the lone surviving individual from earlier plantings established as avenue trees or for timber and matchwood purposes. It was located beside a roadside, adjacent to public buildings.

Field Surveys

Field observations were conducted over the course of the outbreak (25 January to 1 February 2026). The spatial extent of larval webbing, intensity of defoliation, and presence of different developmental stages were recorded through systematic visual surveys and photographic documentation. Incidental human responses (e.g., itching) were documented via reports from site visitors.

Specimen Collection, Identification and studies on natural enemy complex

Larvae and pupae were hand-collected along with the webbed foliage and inflorescences and from surrounding leaf litter. They were carefully transferred to 30 × 25 cm rearing containers lined with tissue paper and provided with fresh host leaves and shoots. Adult moths observed during field surveys were also collected.

Live larvae were reared on fresh *A. triphysa* leaves under ambient laboratory conditions (26–30°C, 60–70% RH) until adult emergence. Adults were identified to species using morphological keys and diagnostic characters consistent with the revised taxonomy of Asian *Atteva* species (Sohn & Wu 2013). Approximately 80 pupae collected during field surveys were incubated to record the natural enemy complex associated with *A. fabriciella*, following methodologies described by Varma (1986, 1991).

Results

Taxonomic Confirmation

Adult specimens emerging from field-collected larvae and pupae were examined using a stereomicroscope, and diagnostic external characters were compared with descriptions and illustrations available in the literature. The specimens conformed to the diagnostic morphology of *Atteva fabriciella* Swederus, including the characteristic orange forewings densely marked with white spots and other morphological features consistent with confirmed Asian records (Sohn & Wu 2013).

Voucher specimens have been preserved in the personal collection of the first author and will be formally deposited in the Forest Insect Collection and Museum of the Kerala Forest Research Institute (KFRI), Peechi, Kerala.

Outbreak Description

High densities of larvae (ranging from 7 to 15 individuals) were observed within extensive silken webs clustered around terminal branches and leaves. Webbed foliage and inflorescences exhibited varying degrees of skeletonization, browning, and silk matting, all indicative of intensive larval feeding activity. . Pupae were found both within the webbing and in dried leaf litter on the ground, and occasionally in bark crevices and nearby buildings, consistent with post-feeding migratory behaviour.

Host Damage and Dermal Responses

The host tree exhibited marked defoliation, with the upper canopy showing the most severe damage.. Field visitors reported intense itching after contact with webbed foliage and larval aggregations, suggesting the possibility of dermatological irritation caused by larval setae or silk-bound particulates. While the mechanism remains undocumented for *A. fabriciella*, similar larval setae in other Lepidoptera cause cutaneous irritation in humans (Gavas et al., 2025). These observations indicate a potential public-health concern when outbreaks occur in publicly accessible or recreational areas.

Natural enemy complex

The pupae of *A. fabriciella* were found parasitized by both dipteran and hymenopteran parasitoids. A total of 80 pupae that were collected during surveys were kept for incubation under laboratory conditions. During the observation period, 23 individuals of *Brachymeria* sp. (Hymenoptera: Chalcididae) emerged as pupal parasitoids, while 7 individuals of tachinid (Diptera: Tachinidae) also emerged from the pupae. flies

Pupal parasitism by *Brachymeria* sp. accounted for 28.75%, whereas tachinid parasitism accounted for 8.75%. These findings suggest that *Brachymeria* sp. is the dominant parasitoid in the natural enemy complex of *A. fabriciella*, potentially contributing significantly to natural regulation of the pest population.

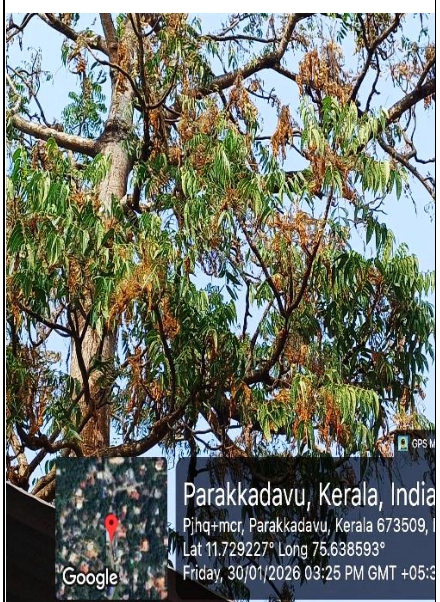
Discussion

Taxonomic and Distribution Context

Atteva fabriciella is a well-recognized species within the family Attevidae, distributed across tropical Asia and associated primarily with simaroubaceous hosts (Sohn & Wu 2013). Recent faunistic work, including records from Cambodia where *A. fabriciella* has been newly documented, underscores the species' widespread Oriental distribution (Na & Bae 2024).

Fig. 1: *Ailanthus triphysa* tree

Fig. 2: *Ailanthus triphysa* tree heavily infested by *Atteva fabriciella*



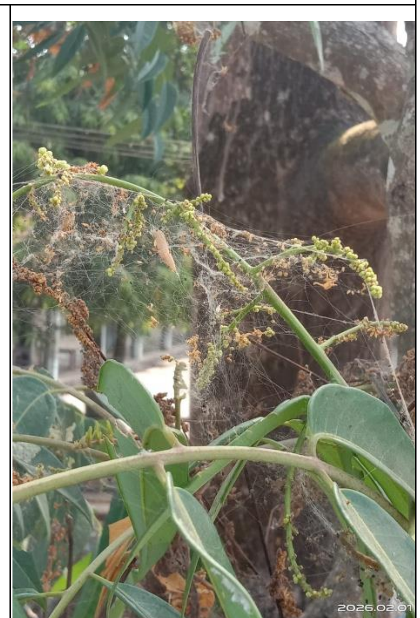


Fig. 3, 4 & 5: Skeletonization and browning of webbed foliage and inflorescences by *Atteva fabriciella*

Fig. 6, 7 & 8 : Larva, Pupa and Adults of *Atteva fabriciella*

Fig. 9 & 10 : Chalcid pupal parasitoid *Brachymeria* **sp. & a new tachinid pupal parasitoid of** *A.fabriciella*

Historical confusion with closely related taxa such as *Atteva wallengreni* has been resolved through morphological revision and neotype designation, confirming the identity of *A. fabriciella* in Indian and broader Asian contexts (Sohn & Wu 2013).

Biology and Host Interactions

Under Indian conditions, the life cycle of *Atteva fabriciella* (Swederus) is relatively short and strongly influenced by temperature, host plant condition, and seasonal factors. Females deposit eggs on tender leaves and shoots of *Ailanthus* species, and the eggs usually hatch within 2–3 days. The larval stage consists of five instars and generally lasts about 14–20 days, during which larvae feed gregariously on young foliage and often spin loose silken webs that bind leaves together. Fully grown larvae pupate within a thin silken cocoon on the host plant or in nearby plant debris. The pupal stage typically lasts 6–12 days under tropical field conditions. Adults emerge soon after pupation, and the adult longevity is usually 4–7 days, during which mating and oviposition occur. Consequently, the complete life cycle from egg to adult is completed in approximately 25–40 days in most parts of India, allowing the insect to produce several overlapping generations throughout the year, particularly in warm regions where *Ailanthus* remains available as a host plant (Varma, 1986 ; Varma, 1991).

Larval gregariousness and communal webbing likely afford protection against predators and microclimatic stress, facilitating rapid population increases under favourable environmental conditions. While outbreaks have been documented previously in plantations and young stands (Varma, 1986), observation on a mature tree highlights the pest's capacity to exploit canopy foliage across age classes. Known host range includes multiple Simaroubaceae species and possibly alternative hosts such as *Quassia indica* in Kerala plantations (Varma, 1986).

Change in insect abundance occurs over time for various reasons such as micro and macro climate change and variation in the availability of food resources (Wolda, 1988). Varma (1996) reported that defoliation by *Atteva fabriciella* lead to reduction in growth increment due to defoliation of tender leaves, forking of tree due to terminal bud damage and loss of seed production due to damage of inflorescence and fruits.

Manimaran et al. (2021) recorded that minimum temperature and wind velocity had significant contribution towards the abundance of Ailanthus webworm (*A. fabriciella*) , which was in conformity with the findings of Varma (1986), who reported that occurrence of defoliators was low during rainy season and more during cool dry season.

Ailanthus triphysa is an important fast-growing plantation tree used for plywood, matchwood, and light timber. Large plantations were established in Kerala, but serious insect pest outbreaks were observed mostly in nurseries and young plantations. The present record of *A. fabriciella* outbreak on isolated mature trees points to the high adaptability of the pest.

Natural enemy complex

In his comprehensive study on s*easonal incidence of A.fabriciella in plantations of Ailanthus triphysa, Varma (1986) observed that a* chalcid, *Brachymeria hime attevae* was *the* only parasite found associated with *A. fabriciella.* He could record only few instances of parasitism during the study and couldn't ascertain the level of parasitization. But Varma (1991) in his follow up studies states that the pupae of *A.fabriciella* was found parasitized by *Brachymeria hime attevae* during the months of January, February and May in 1988 and over 20% parasitism was recorded. But he cautiously points out that during the said period the pest incidence was not very high.

In our present study, chalcid parasitoid *Brachymeria* spp. and an unidentified tachinid pupal parasitoid were recorded from the field collected *A. fabriciella* pupae. The extent of parasitism exhibited by Brachymeria sp. (28.75%) is similar to the findings of Varma (1991), but the pest incidence was high during the preiod. The new record of a tachinid pupal parasitoid during the present study, points to the need of further studies on natural enemy complex of *A.fabriciella.* This points to the high potential of these parasites as a great pest mortality factor in field level for the natural regulation of its populations.

Management Implications

Given the ability of *A. fabriciella* to defoliate trees and cause public discomfort through dermal interaction, integrated monitoring programs and early detection are recommended, especially in urban and recreational landscapes where human contact is likely. Future research should investigate environmental triggers of outbreaks, natural enemy dynamics, and control measures that minimize ecological and human health impacts.

Conclusions

This study documents a significant infestation of *Atteva fabriciella* on a mature *Ailanthus triphysa* in Kerala, providing new insights into the species' outbreak potential and its interactions with mature host trees.. The inclusion of updated taxonomic information, global distribution records, and observations of dermal irritation enhances understanding of the ecological and public-health relevance of this pest. The role of the natural enemy complex warrants further investigation to evaluate their potential for incorporation into sustainable, eco-friendly management programs.

Acknowledgment

The immense help in confirming the host plant identity by Dr. V. Jamaludheen, Professor (Silviculture & Agroforestry), ICAR-AICRP on Agroforestry, College of Forestry, Kerala Agricultural University, Vellanikkara is kindly acknowledged.

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MS Received on 1st February, 2026
MS Accepted on 1st March, 2026

DOI: 10.55278/HXIO9937

A Preliminary Checklist of Acrididae (Insecta: Orthoptera) fauna from Garbhanga Reserve Forest, Assam

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Abstract

The family Acrididae (Orthoptera: Acridoidea) comprises ecologically significant grasshoppers that play vital roles in nutrient cycling, herbivory, and food-web dynamics. However, their diversity remains insufficiently documented across many protected and semi-protected forest habitats of Assam. This study provides the first preliminary inventory of Acrididae from the Garbhanga Reserve Forest, part of the ecologically important Rani–Garbhanga landscape in Assam. A total of 17 species, representing 13 genera and seven subfamilies, are recorded for the first time from this reserve. The findings offer essential baseline data to support future taxonomic studies, biodiversity monitoring, and conservation planning within this rapidly changing landscape.

Key words: Acrididae, Garbhanga Reserve Forest, Orthoptera, Assam

Introduction

The family Acrididae (order Orthoptera, superfamily Acridoidea) comprises grasshoppers, including locusts, and is widely distributed across India. Acridoidea superfamily contains 11 families, of which Acrididae is the largest and most diverse, encompassing approximately 6,889 species in 28 subfamilies worldwide (Cigliano *et al*., 2025). In India, 366 Acrididae species representing 16 subfamilies are currently recognised (Chand *et al*., 2025).

Grasshoppers and locusts (Acrididae) are economically important orthopterans that infest both cultivated and non-cultivated vegetation. The family name Acrididae was first used by Krauss (1890), although the priority for family-group names based on *Acrida* dates to Acridina proposed by MacLeay (1821), (Cigliano *et al*., 2025). Globally, these insects can cause substantial damage to agricultural crops, pastures, and forest vegetation (Joshi *et al*., 1999). Their diet is primarily graminivorous, feeding on a variety of grasses and sedges (Mulkern, 1967).

Members of Acrididae can be identified by a series of morphological features: a head with a broad frontal ridge; fastigial furrow absent; fastigial foveolae present or absent; a median depression; a pronotal dorsum that is short and variably shaped, usually bearing median and lateral carinae; the prosternal tubercle present or absent; tegmina and wings fully developed, reduced, or absent; tympanum usually present; and the lower basal lobe of the hind femur shorter than or equal to the upper (Kirby, 1914).

Assam contains approximately 300 reserve forests, among which Garbhanga Reserve Forest is one of the largest (Assam Forest Department, 2011–2012). Situated immediately south of Guwahati, Garbhanga is ecologically connected to the adjacent Rani Reserve Forest; together they form the Rani–Garbhanga landscape (Devi *et al*., 2012), one of the most extensive reserve-forest networks in the state. Garbhanga comprises a series of undulating hills ranging from 80 to 670 m a.s.l. (Mahananda *et al*., 2023). Despite proximity to a rapidly expanding metropolitan area, the forest harbours rich biodiversity: 128 bird species (Lahkar *et al*., 2010), 54 butterfly species (Modak *et al*., 2018), and 29 swallowtail species/subspecies (Barua *et al*., 2004) have been reported so far. More recently, Mahananda *et al*. (2023) documented 254 butterfly species (6 families), 29 amphibians (7 families), 64 reptiles (12 families), 307 birds (68 families), and 31 mammals (19 families) from the area.

The varied topography, microhabitats, and forest types of Garbhanga provide niches for a broad spectrum of taxa. However, increasing anthropogenic pressures—such as land-use change, infrastructure development, and habitat degradation—threaten the integrity of this ecosystem. Given their dominance and ecological importance within Orthoptera, Acrididae serve as effective bioindicators of habitat quality, vegetation structure, and climatic variability. They contribute to primary herbivory, nutrient cycling, and the food web as prey for birds, reptiles, and small mammals. Documenting the Acrididae fauna of Garbhanga is therefore crucial both for understanding local biodiversity and for assessing ecological health. This preliminary study provides baseline information on Acrididae diversity in Garbhanga Reserve Forest, supporting future taxonomic work, biodiversity monitoring, and conservation planning.

Materials and Method

Study Area

Garbhanga Reserve Forest (Figure 1) covers approximately 110 km² and is geographically situated between 91°35.406′ E to 91°47.517′ E longitudes and 25°56.528′ N to 26°06.584′ N latitudes (Tamuli, 2018; Barua *et al*., 2004). The forest lies along the southern bank of the Brahmaputra River and borders the greater Guwahati metropolitan region (Saikia & Saikia, 2015). It occupies a unique ecological position—bounded by the Meghalaya hill ranges to the east and south, Rani Reserve Forest to the west, and Guwahati city together with Deepor Beel Wildlife Sanctuary to the north.

The elevation ranges from 100–200 m a.s.l., forming a landscape of undulating hills (Barua *et al*., 2004). The region experiences four distinct seasons—pre-monsoon, monsoon, post-monsoon, and winter (Lahkar *et al*., 2010). This physiographic and climatic variability supports diverse vegetation types and microhabitats suitable for Orthopteran fauna.

Sampling and Specimen Collection:

Field surveys were conducted during 2025. Grasshoppers were collected using:

- Sweep-net sampling: repeated sweeping across grasses, shrubs, and low vegetation;
- Hand collection: manually picking larger or less agile individuals using fingers or fine forceps.

Specimens were euthanized using ethyl acetate in killing jars and transferred into insect envelopes for temporary field storage. All samples were transported to the laboratory for further processing.

In the laboratory, specimens were pinned, labeled, dried, and preserved following standard entomological procedures. Geographic coordinates for each sampling site were recorded using a Garmin Oregon 550 GPS unit.

Detailed morphological examination was performed under stereo zoom binocular microscopes, using standard identification keys and literature. All confirmed specimens were deposited in the National Zoological Collection, North Eastern Regional Centre, Zoological Survey of India (ZSI), Shillong.

Collection Localities:

Specimens were collected from four localities within Garbhanga Reserve Forest (Figure 3):

Sites	Latitude	Longitude	Altitude
1.Garbhanga	26.027776°N	91.72279°E	43 m
2.Lokhra	26.093346°N	91.748942°E	51 m
3.Borpani	26.093293°N	91.749024°E	50 m
4.Jaluk Paham	25.978704°N	91.707896°E	72 m

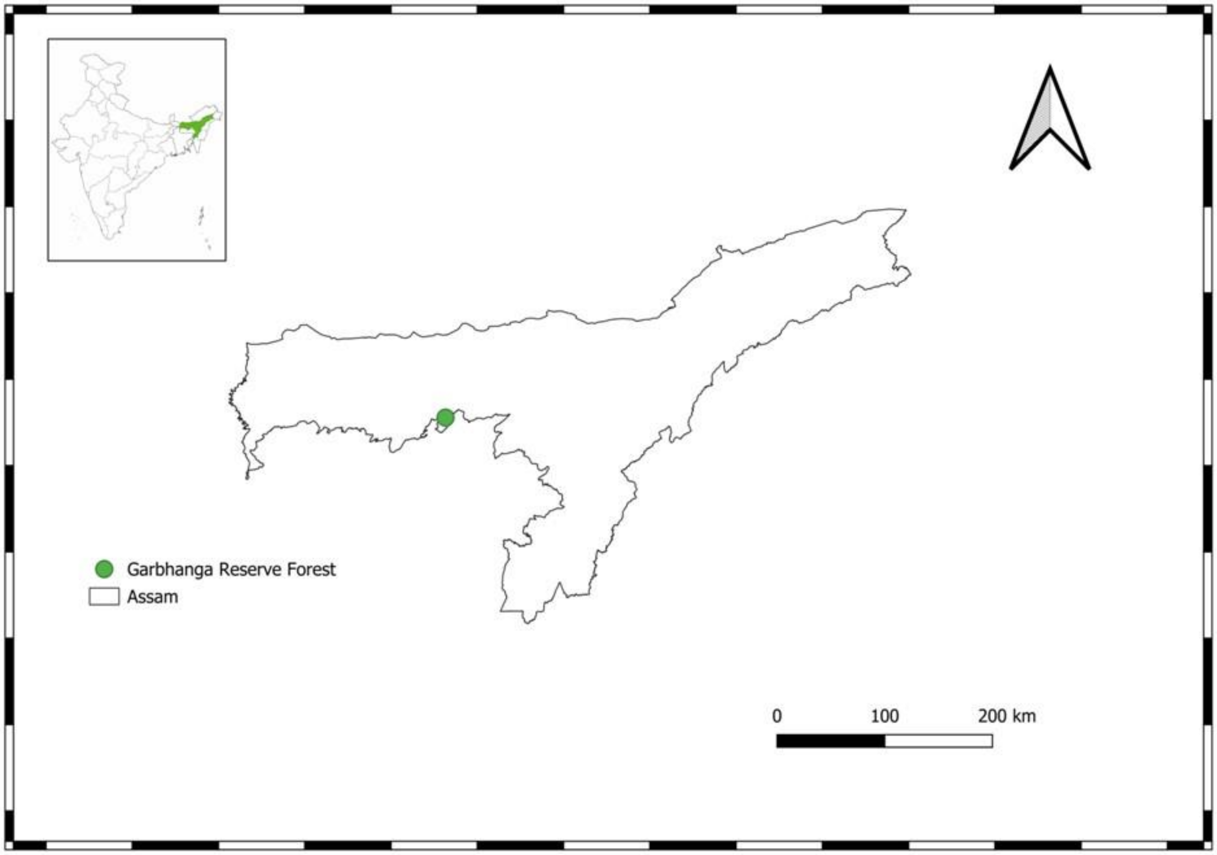

Fig. 1: Map showing location of Garbhanga Reserve Forest in Assam.

Result and Discussion

A number of taxonomic studies on the Acrididae diversity of Assam have been carried out over more than a century, beginning with the early works of Kirby (1914) and Hancock (1915), followed by Chopard (1969). Subsequent contributions include Bhowmik *et al.* (1990), Senthilkumar *et al.* (2006), Senthilkumar (2010), and Usmani and Khan (2010). Further studies were undertaken by Senthilkumar and Borthakur (2013) and Chowdhury *et al.* (2015). In 2016, important additions were made by Dutta *et al.* (2016), Khan *et al.* (2016), and Khan and Usmani (2016), followed by Borkakati *et al.* (2018). More recent works include Gupta and Chandra (2019), Gupta *et al.* (2021),Bhowmick *et al.* (2021), Mukherjee *et al.* (2022) and Choudhury and Hazarika (2022). This was followed by Das *et al.* (2023), Muchahary *et al.* (2023), and Ahmed and Islam (2023). The most recent contributions are by Basumatary *et al.* (2024), Das *et al.* (2024), and Narjari and Narzari (2024). However, the study of diversity of this group is very limited in the protected areas of Assam.

Senthilkumar *et al.* (2006) reported 7 species of Acridiae from the Gibbon Wildlife Sanctuary. In 2010, Senthilkumar reported 19 species of Acrididae from the Kaziranga National Park of Assam. Mukherjee *et al.* (2022) while preparing the first faunal inventory of the newly declared Raimona National Park reported 5 species of Acrididae. Apart from these studies no major work has been done on Acrididae fauna of the protected areas of Assam.

Table 1: List of Acrididae species reported from Garbhanga Reserve Forest

Sl. No.	Subfamily	Genus	Species	Habitat
1	Acridinae	*Acrida*	*Acrida exaltata* (Walker, 1859)	Grassland
2		*Phlaeoba*	*Phlaeoba infumata* Brunner Von Wattenwyl, 1893	Shrubs
3			*Phlaeoba antennata* Brunner Von Wattenwyl, 1893	Grassland, Shrubs
4	Oxyinae	*Oxya*	*Oxya hyla* Serville, 1831	Grassland
5			*Oxya japonica japonica* (Thunberg, 1824)	Grassland
6			*Oxya velox* (Fabricius, 1787)	Grassland
7		*Pseudoxya*	*Pseudoxya diminuta* (Walker, 1871)	Grassland, Shrubs
8	Oedipodinae	*Dittopternis*	*Dittopternis venusta* (Walker, 1870)	Shrubs, sandy soil
9		*Trilophida*	*Trilophida annulata* (Thunberg, 1815)	Sandy soil
10		*Ceracris*	*Ceracris nigricornis nigricornis* Walker, 1870	Shrubs
11	Catantopinae	*Diabolocatantops*	*Diabolocatantops innotabilis* (Walker, 1870)	Shrubs
12		*Xenocatantops*	*Xenocatantops humilis* (Serville, 1838)	Shrubs
13	Coptacrinae	*Eucoptacra*	*Eucoptacra binghami* Uvarov, 1921	Shrubs
14			*Eucoptacra praemorsa* (Stal, 1861)	Shrubs
15		*Apalacris*	*Apalacris varicornis* Walker, 1870	Shrubs
16	Spathosterninae	*Spathosternum*	*Spathosternum prasiniferum prasiniferum* (Walker, 1871)	Grassland, Shrubs
17	Eyprepocnemidinae	*Choroedocus*	*Choroedocus robustus* (Serville, 1838)	Shrubs

Table 2: Acrididae fauna in different localities of Garbhanga Reserve Forest

Sl. No.	Name of Species	Garbhanga	Lokhra	Borpani	Jaluk Paham
1	*Acrida exaltata* (Walker, 1859)			1♂	
2	*Phlaeoba infumata* Brunner Von Wattenwyl, 1893	3♂, 2♀	3♂, 5♀	4♂, 2♀	2♂, 4♀
3	*Phlaeoba antennata* Brunner Von Wattenwyl, 1893	2♂, 4♀	3♂, 5♀	3♂, 3♀	2♂, 3♀
4	*Oxya hyla* Serville, 1831	1♀	1♀	3♀	1♀
5	*Oxya japonica japonica* (Thunberg, 1824)	3♂, 1♀	1♂, 1♀	2♂, 1♀	1♂
6	*Oxya velox* (Fabricius, 1787)	1♂, 2♀	1♂, 1♀	1♀	
7	*Pseudoxya diminuta* (Walker, 1871)			1♂, 1♀	3♂, 2♀
8	*Dittopternis venusta* (Walker, 1870)		1♂	1 F	1♂
9	*Trilophida annulata* (Thunberg, 1815)	3♂, 3♀	2♂, 2♀	2♂, 1♀	1♀
10	*Ceracris nigricornis nigricornis* Walker, 1870	1♀	1♂, 1♀	2♂, 2♀	2♂
11	*Diabolocatantops innotabilis* (Walker, 1870)			2♀	1♀
12	*Xenocatantops humilis* (Serville, 1838)	2♂, 5♀	3♂, 4♀	1♂, 3♀	3♂, 3♀
13	*Eucoptacra binghami* Uvarov, 1921		1♀		1♂, 2♀
14	*Eucoptacra praemorsa* (Stal, 1861)	1♀	1♀	3♂	1♂, 1♀
15	*Apalacris varicornis* Walker, 1870				1♂
16	*Spathosternum prasiniferum prasiniferum* (Walker, 1871)	3♂, 1♀	2♀	2♂, 2♀	
17	*Choroedocus robustus* (Serville, 1838)				1♂

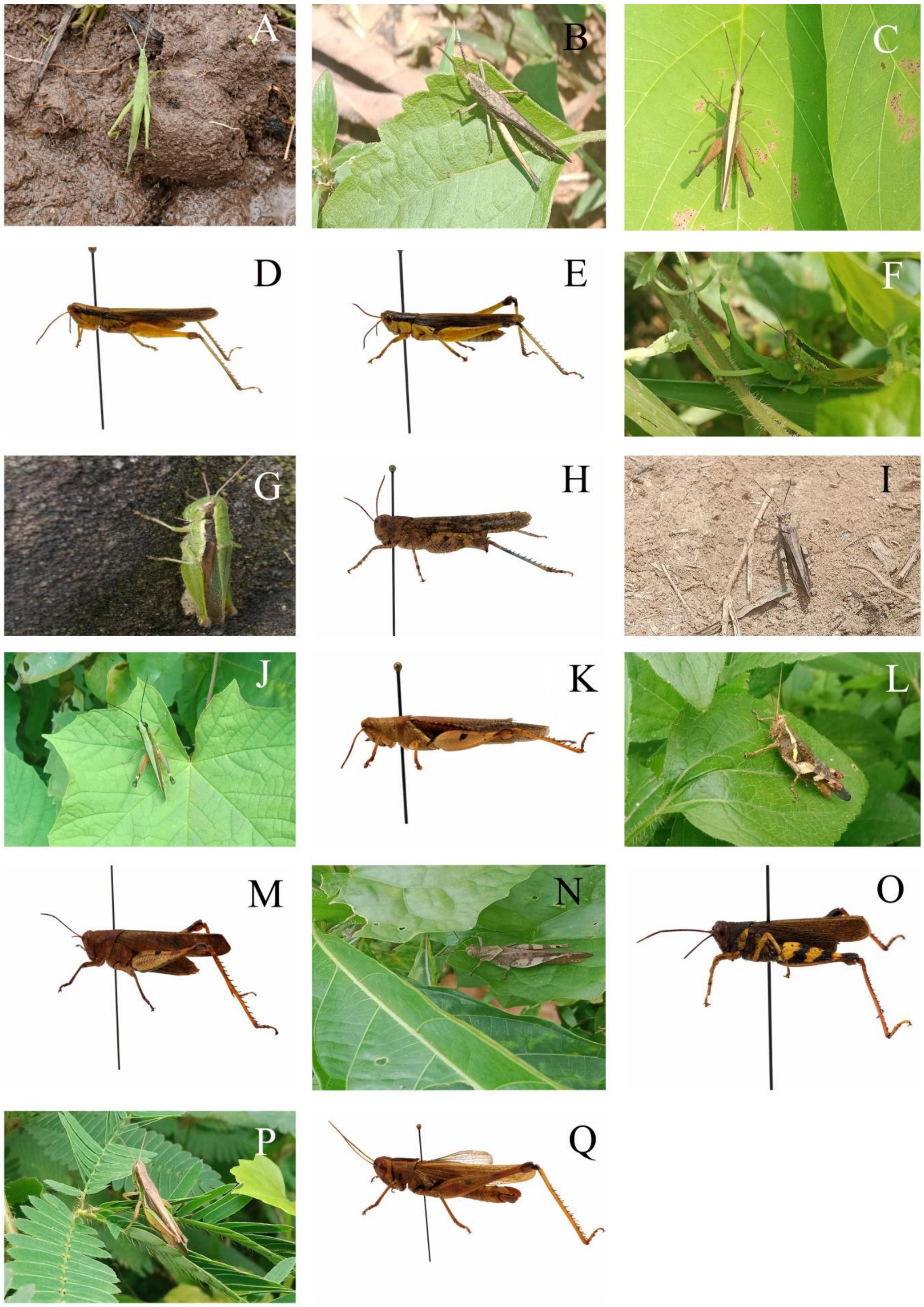

Fig. 2: Acrididae species recorded from Garbhanga Reserve Forest. A- *Acrida exaltata*, B- *Phlaeoba infumata*, C- *Phlaeoba antennata*, D- *Oxya hyla*, E-*Oxya japonica japonica*, F- *Oxya velox*, G-*Pseudoxya diminuta*, H- *Dittopternis venusta*, I- *Trilophidia annulata*, J- *Ceracris nigricronis nigricornis*, K- *Diabolocatantops innotabilis*, L- *Xenocatantops humilis* M-*Eucoptacra binghami*, N- *Eucoptacra praemorsa*, O- *Apalacris varicornis*, P- *Spathosternum prasiniferum prasiniferum*, Q- *Choroedocus robustus*.

Fig. 3: Collection sites of the Reserve Forest. A- Borpani, B- Jaluk Paham, C- Garbhanga, D- Lokhra

This preliminary investigation documents 17 species of Acrididae, representing 13 genera and seven subfamilies, from the Garbhanga Reserve Forest—all reported for the first time from this protected area **(Table 1 and Table 2; Fig. 2)**. Among the recorded taxa, *Phlaeoba infumata* Brunner von Wattenwyl, 1893, *P. antennata* Brunner von Wattenwyl, 1893, and *Xenocatantops humilis* (Serville, 1838) were particularly abundant, predominantly inhabiting grassland and shrub-dominated microhabitats.

Although Assam has a long history of Acrididae research, very few studies have focused on protected areas, leaving substantial gaps in the faunal understanding of these ecosystems. The present study fills part of this gap by providing the first baseline dataset for Acrididae diversity within the ecologically important Garbhanga Reserve Forest.

Given the forest's heterogeneous landscape and its conservation significance, it is highly probable that continued and season-wise surveys will reveal additional Acrididae species, including rare or habitat-specialist taxa. Such comprehensive documentation will be crucial for future biodiversity assessments, monitoring ecological health, and formulating conservation strategies for Orthopteran fauna in Assam's protected landscapes.

Acknowledgement: We would like to thank the Assam Forest Department and Assam state biodiversity board for the permission to carry out the study. We are thankful to the Head of The Department of Zoology, Girijananda Chowdhury University for supporting us. We would also like to thank the local residents near Garbhanga Reserve Forest for helping us during the survey and collection of specimens. The corresponding author would like to thank the Director, Zoological Survey of India and the Officer-in-Charge of Northeastern Regional Centre, Zoological Survey of India for providing necessary facilities to carry out this work.

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MS Received on 03rd February, 2026
MS Accepted on 03rd March, 2026

DOI: 10.55278/YXSN9808

Bridging the Wallacean Shortfall: New Distributional Records of Two Elaterid Beetles from Maharashtra, India

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Abstract

The present study documents new geographic records of *Lanelater fuscipes* (Fabricius, 1775) and *Priopus vafer* (Erichson, 1841) (Coleoptera: Elateridae) from Maharashtra, India. These findings extend the known distributional ranges of both species and contribute valuable data toward addressing the Wallacean shortfall an important gap in our understanding of species' spatial distributions. The newly recorded occurrences enhance current biogeographic knowledge of the family Elateridae in India and provide baseline information useful for future ecological, taxonomic, and pest-management research.

Key words: Wireworm, Elaterinae, Agrypninae, pest, morphology, male genitalia.

Introduction

The beetle family Elateridae Leach, 1815 commonly known as click beetles or wireworms represents one of the most diverse and ecologically significant lineages within Coleoptera. Members of this family possess a distinctive prosternal–mesosternal clicking mechanism. Due to this they produce a sudden snapping sound and propel themselves into the air when overturned. This mechanism serves important defensive and evolutionary functions (Costa *et al*., 2010). Globally, the Elateridae comprises approximately 11,000 described species across about 600 genera and 20 subfamilies (Bouchard *et al*., 2017; Szabó *et al*., 2022). In India, nearly 835 species in 113 genera distributed across eight subfamilies have been documented (Tara, 2021). Despite this diversity, Indian elaterid fauna remains poorly explored, with several biogeographic regions still under-surveyed.

Larvae of Elateridae exhibit diverse ecological roles and may broadly be categorised into root-feeding soil pests and predatory or wood-boring forms. Root-feeding wireworms are of particular agricultural importance as they damage seeds, roots, tubers, and underground stems. Adults are typically nocturnal, feeding on nectar and pollen, while some species are phytophagous;

during the day, they remain concealed under soil debris or organic matter (Seal 1991; Seal *et al*. 1992).

Among subterranean pests, elaterid larvae are especially destructive. Their feeding activity affects crops such as maize, groundnut, sugarcane, potato, wheat, sorghum, onion, ginger, chilli, carrot, and sugar beet, damaging vital subterranean tissues and often facilitating secondary infections by pathogenic fungi and bacteria (Anandmurthy *et al.,* 2025). Reported crop damage ranges from 20.93% to 37.16%, with severity varying by crop growth stage early seedling stages in maize and sugarcane, germination and pod-filling in groundnut, and the vegetative stage in ginger and onion (Anandmurthy *et al.,* 2025).

Lanelater fuscipes (Fabricius, 1775) is a widely distributed tropical insect whose grubs burrow into roots and tubers.Their attack causes stunted growth and yield losses ranging from 10–30% (Anandmurthy et al. 2025) under severe infestations. This attack has been reported in various cultivated crops, including sugarcane, potato, groundnut, maize, rice, and several grasses and ornamental plants. It is generally considered a minor pest. But its localised outbreaks may necessitate integrated management approaches, including cultural practices and the application of entomopathogenic organisms. *Priopus vafer* (Erichson, 1841) is a small click beetle of Oriental origin belonging to the subfamily Elaterinae. Less data is available on this particular wireworm from India.

Significant knowledge gaps about species richness and diversity still exist. Even though Indian Elateridae documentation has advanced in the recent past... Notably, both the Linnean shortfall (incomplete taxonomic knowledge) and the Wallacean shortfall (insufficient understanding of species' geographic distributions) are evident in this group (Vergara-Asenjo *et al*., 2023). Knowledge gaps are pronounced in hyperdiverse groups like Elateridae due to inadequate regional surveys. Addressing these gaps through updated species records is essential. This improves biogeographic understanding, refines pest risk assessments, and strengthens biodiversity documentation. The present study contributes to reducing the Wallacean shortfall by providing new geographic records of two elaterid species from Maharashtra. This will enhance our current knowledge of their distribution and ecological significance in the region.

Materials and Methods

The Specimens were collected at night using a light trap set in the field. The collected specimens were euthanised by ethyl acetate vapours and preserved in 70% alcohol. The specimens were brought to the laboratory and were relaxed. Morphological examination was carried out using a Leica S9i stereo zoom microscope equipped with photographic documentation. The specimens have been confirmed using Naz *et al.* (2012) and Platia (2015). Identification was based on external morphology and male and female genitalia by referring to standard literature. All examined material has been deposited in the National Repository of the Zoological Survey of India, Western Regional Centre, Pune, Maharashtra, India.

Results and Discussion

Systematic account

Family: Elateridae Leach, 1815
Subfamily: Agrypninae Candèze, 1857
Tribe: Pseudomelanactini Arnett, 1967
Genus: *Lanelate*r Arnett, 1952
Lanelater fuscipes (Fabricius, 1775): 211 (*Elater*) (Fig. 1)

Material examined. 01 male, Yawal, Jalgaon, Maharashtra, 1.vii.2019, coll. A.S. Kalawate, (ZSI-WRC-Ent. 1/5300); 01 male, ZSI, WRC campus, Akurdi, Pune, Maharashtra, 1.xii.2022, coll Vidya, (ZSI-WRC-Ent. 1/5306); 01 female, Patan, Satara, Maharashtra, 12.vii.2017, coll. A.S. Kalawate, (ZSI-WRC-Ent. 1/5299).

Diagnostic character. Adult male (Figure 1), 33 mm in length. Elongate-parallel, blackish-brown with rusty lateral margins. Pronotum strongly punctate with sparse yellowish recumbent pubescence, posterior angles sharply acute and extending posteriorly beside elytra bases; antennae serrate. Scutellum small and transverse; elytra striate with deep fine punctures, they are widest in the middle and have separately rounded tips; legs elongate, hind tarsi with four ventral pads. The male genitalia have median lobe that is widest in the middle and tapers to a narrow, cylindrical apex.

Adult female (Figure 1), length 38 mm. Similar to the male but larger and sturdier, with a darker blackish-brown body and fewer reddish tones. The pronotum is more rounded on top, and the lateral carinae are straighter. The antennae are slightly shorter than the body. The elytra are more parallel-sided, and the abdomen is somewhat broader and sturdier.

Distribution. India: Karnataka, New Delhi, Sikkim, Tamil Nadu, Uttar Pradesh, West Bengal (Kharel et al. 2020; Kotru et al. 2025) Maharashtra (present study). Elsewhere. China, Madagascar, Nepal, Pakistan, Sri Lanka; Afrotropical Region, Oriental Region, Reunion Island (Cate 2007; Kotru et al. 2025).

Remark. New record for Maharashtra.

Subfamily: Elaterinae Leach, 1815
Tribe: Melanotini Candèze, 1859
Genus: *Priopus* Laporte, 1840
Priopus vafer (Erichson, 1841): 108 (*Cratonychus*) (Fig. 2)

Material examined. 01 male, Yawal, Jalgaon, Maharashtra, 1.vii.2019, coll. A.S. Kalawate, (ZSI-WRC-Ent 1/ 5305).

Diagnostic characters. Adult male (Figure 2), length 11.40 mm. The body is elongated, parallel-sided, and moderately flattened. It has a general colouration of dark brown to blackish-brown with a dull metallic shine on the elytra. The antennae and legs are lighter and covered with yellowish hairs. The pronotum is bell-shaped, measuring 1.3 times longer than wide. Its surface is finely punctured with sparse pale yellowish hairs. The frons is slightly raised, and the eyes are moderately prominent. The antennae are serrated from antennomere III onward. They are moderately long and do not extend beyond the posterior angles of the pronotum; antennomeres III–X are triangular, while XI is elongated and oval. The scutellum is small, transverse, and nearly square. The elytra are elongated, parallel-sided, and striated. The striae are deep and have fine punctures, while the interstriae are flat to slightly raised. The prothorax is loosely connected to the mesothorax, creating a functional clicking mechanism. The legs are slender and long. The male genitalia have a median lobe elongated, cylindrical, and tapers at the tip. Paramere structures are symmetrical, elongated, and slightly curve inward at the tip, surrounding the median lobe.

Distribution. India: Odisha, Sikkim, West Bengal (Platia & Schimmel 1995; Platia 2015), Maharashtra (present study). Elsewhere. China, Pakistan (Platia, 2015), Indonesia, Laos, Malaysia, Myanmar, Sri Lanka, Thailand, Vietnam, (Anonymous 2026; Platia & Schimmel 1995).

Remark. New record for Maharashtra.

The present study documents new distributional records of *L. fuscipes* and *P. vafer* from Maharashtra. This study helps in expanding the known geographic range of both species within India. Detailed morphological examination, including diagnostic external characters and male and female genitalia, confirmed the identity of all specimens. These findings contribute valuable data toward addressing the Wallacean shortfall by improving knowledge of regional elaterid diversity and species occurrence. The new records not only strengthen the biogeographic understanding of the families Agrypninae and Elaterinae in the Indian subcontinent but also provide important baseline information for future studies.

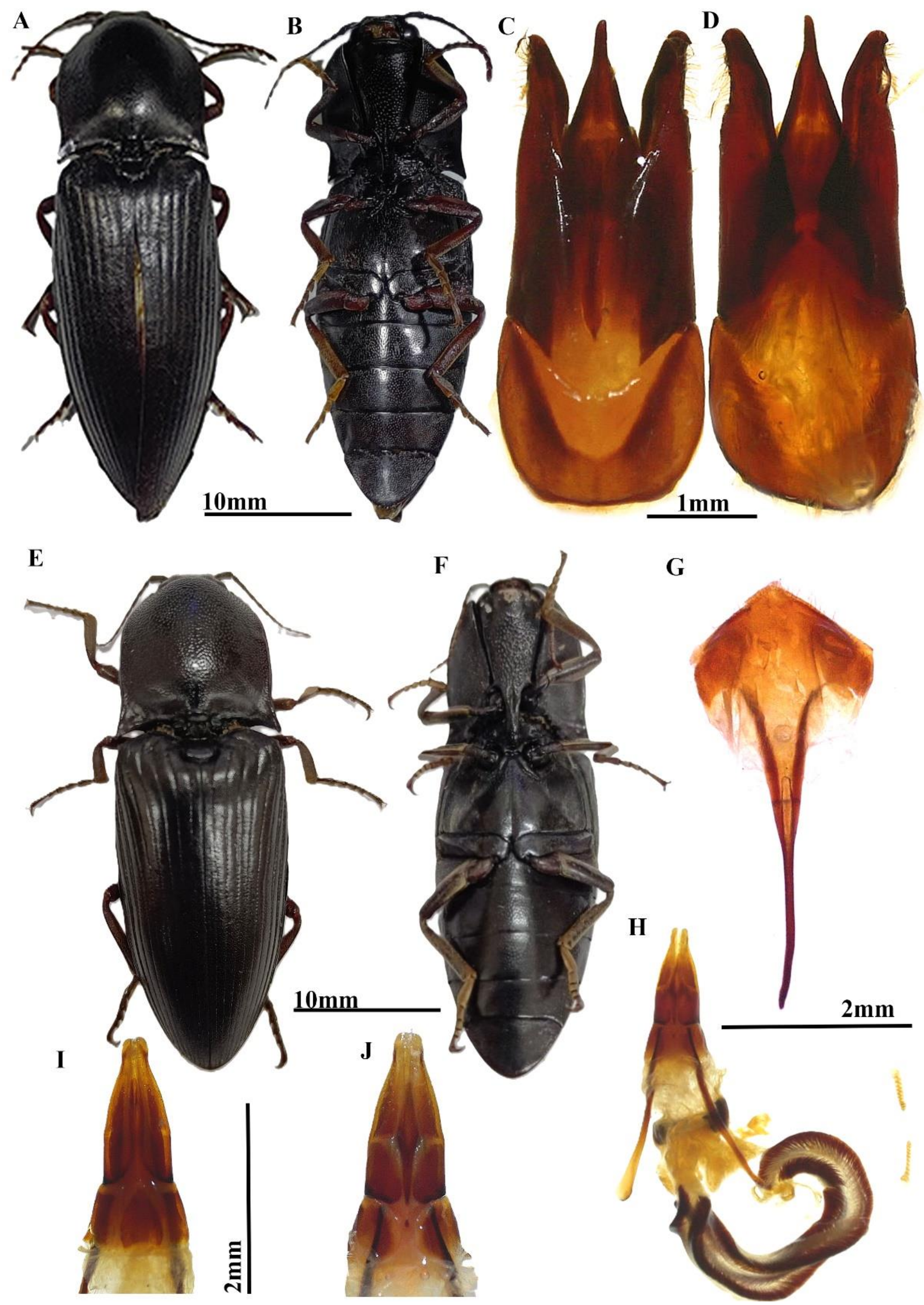

Fig. 1. ***Lanelater fuscipes*** **(Fabricius, 1775) A–D ♂: A, Habitus, dorsal view; B, Habitus, ventral view; C, Genitalia, dorsal view; D, Genitalia ventral view; E–J♀: Habitus, dorsal view, F, Habitus, ventral view, G, sternite, H, female genitalia, I, close ventral view of female genitalia, J, close dorsal view of female genitalia.**

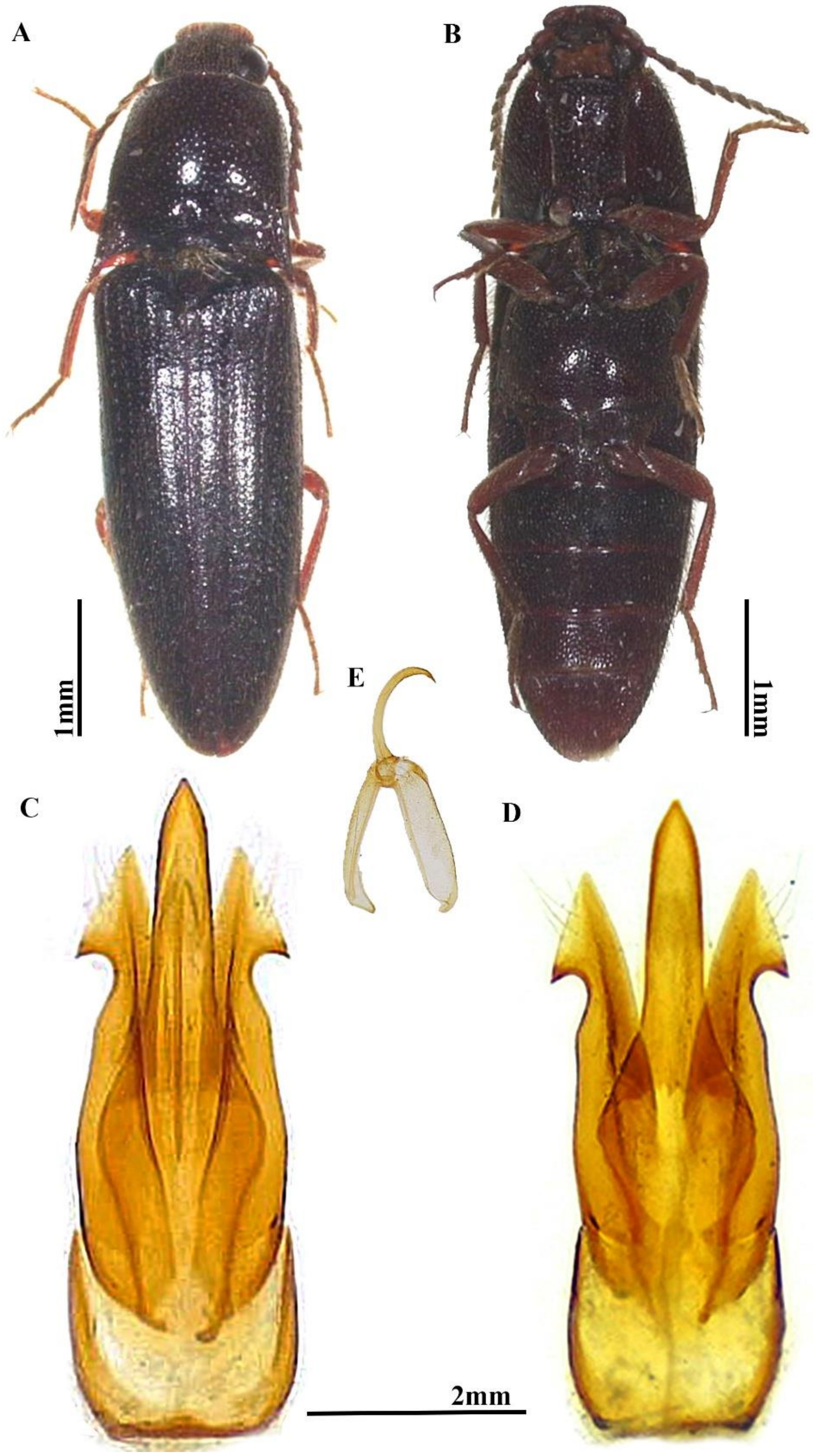

Figure 2. ***Priopus vafer*** **(Erichson, 1841) ♂. A, Habitus, dorsal view; B, Habitus, ventral view; C, Male genitalia dorsal view, D. Male genitalia ventral view); D, subapical hook of parameres.**

Acknowledgements

We would like to thank the Director, Zoological Survey of India, Kolkata and the Officer-in-Charge, ZSI-WRC, Pune, for their support and access to research facilities. The authors are grateful for the comments and suggestions from the reviewers. We thank Alexander S. Prosvirov, Department of Entomology, Faculty of Biology, Lomonosov Moscow State University, Leninskie gory 1-12, 119234 Moscow, Russia for identification confirmation. Thanks are due to the Maharashtra Forest Department for permission, permit and logistic support during the survey.

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MS Received on 03rd February, 2026
MS Accepted on 3rd March, 2026

DOI: 10.55278/OISX3632

Comparative Evaluation of Cotton, Bhendi and Brinjal as Post Plants for the Mass Rearing of *Aphis gossypii* Glover under Controlled Laboratory Conditions

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Abstract

The influence of host plant species on the developmental and reproductive performance of *Aphis gossypii* was assessed under laboratory conditions using cotton (*Gossypium hirsutum*), brinjal (*Solanum melongena*) and bhendi (*Abelmoschus esculentus*) as test hosts. Significant variation was observed in several biological parameters across the three hosts. The total nymphal duration was shortest on cotton (6.87 days) and prolonged on brinjal (8.07 days) and bhendi (8.73 days). The pre-reproductive period was also significantly shorter on cotton, whereas the reproductive period and adult longevity did not differ markedly among hosts. Fecundity varied significantly, with the highest reproductive output recorded on cotton (25.13 nymphs/female), followed by brinjal (23.13 nymphs/female) and bhendi (17.87 nymphs/female). The shorter developmental period and enhanced fecundity on cotton indicate superior host suitability and greater population build-up potential. The findings demonstrate that host plant species play a critical role in determining the growth and reproductive success of *A. gossypii*. Overall, the results clearly demonstrate that host plant species play a crucial role in shaping the growth and reproductive success of *A. gossypii*, with cotton identified as the most suitable host for efficient laboratory mass rearing.

Keywords: *A. gossypii, biological parameters, fecundity and mass rearing.*

Introduction

The cotton aphid, *Aphis gossypii* Glover (Hemiptera: Aphididae), is a cosmopolitan, highly destructive polyphagous insect pest (Uma *et al.,* 2023). It has an extremely wide host range, attacking more than 900 plant species across the globe (Singh & Singh, 2015). Nymphs and adults cause substantial damage to the stems and leaves of plants using their piercing and sucking mouthparts. This feeding behaviour leads to significant nutritional loss in crops, affecting their overall productivity. In addition, the cotton aphid excretes honeydew and transmits viral diseases. It has been reported to transmit over thirty viruses to crops worldwide (Ebert & Cartwright, 1997).

Biologically, *A. gossypii* exhibits remarkable phenotypic plasticity and an incredibly rapid reproductive capacity. In tropical and subtropical climates, the species predominantly undergoes an anholocyclic life cycle, reproducing continuously through parthenogenesis without a sexual phase (Saha *et al*., 2016). The harm of aphids to crops is mainly caused by parthenogenesis induced rapid population expansion (Lu *et al*., 2024).

Although several eco-friendly management strategies are available, chemical insecticides remain the primary approach for controlling insect pests, including *A. gossypii*. However, their indiscriminate application can lead to sublethal effects and accelerate the development of resistance (Gul *et al.,* 2025). Cotton aphids have evolved resistance to a wide range of insecticides, including organochlorines, organophosphates, carbamates, pyrethroids and neonicotinoids (IRAC, n.d.). The aphids are difficult to control because of their high multiplication rate and ability to evolve resistance to many insecticide classes (Ullah *et al*. 2023). Around the world, so many studies are being conducted to find an effective non-chemical method to manage this serious pest. Hence, there is a need for mass rearing of *A. gossypii* in controlled conditions to conduct experiments without any environmental limitations. For that, the host should be nutritional, quickly produced and less expensive. Compared to the artificial diet, insects reared on a natural host are always advantageous, as it provides complete nutrition for the growth and development of the insect (Uma *et al.,* 2023). Therefore, the study aims to evaluate the biology and reproduction parameters of *A. gossypii* on three important natural hosts to reveal the best host that suits for mass rearing in laboratory conditions.

Materials and Methods

The study was conducted in 2024 at the Department of Entomology, Faculty of Agriculture, Annamalai University, Chidambaram, Tamil Nadu, India. The entire experiment was carried out under standardized laboratory conditions to ensure uniformity across host plant treatments.

Insect culture

A laboratory culture of *A. gossypii* was established from field-collected individuals obtained from unsprayed cotton plants. The culture was maintained separately on pesticide-free, healthy seedlings cotton, bhendi and brinjal under laboratory conditions (25 ± 2°C, 70 ± 5% RH and 12:12 h light: dark photoperiod). Fresh plants were provided periodically to maintain healthy aphid colonies. Prior to initiating the experiment, aphids were reared for two generations on their respective hosts to eliminate field-acquired variability and acclimatize them to laboratory conditions.

Maintenance of host plants

Seeds of Cotton (MCU 13), Bhendi (Arka Anamika) and Brinjal (Annamalai) were sown in earthen pots (30 cm diameter) containing sterilized potting mixture a 2:1:1 ratio (soil: sand: FYM). Plants were raised inside an insect rearing room. No insecticide was applied during the experimental period. Plants aged 25–30 days with tender, fully expanded leaves were selected for biological observations to ensure uniform feeding substrate across treatments.

Experimental design

The biology of *A. gossypii* was studied separately on the three host plants. Five gravid females were transferred carefully onto the underside of tender leaves using a fine camel hair brush and confined with clip cages. After 12 hours, newly produced nymphs were obtained.

All aphids except one neonate were removed, ensuring that each single nymph constituted one replication.

Each host plant treatment consisted of 15 replications. These nymphs were observed every 24 hr till their death. The developmental stages were monitored daily, and moulting to the subsequent stage was confirmed by the presence of shed exuviae.

After adult emergence, observations were recorded on pre-reproductive period, reproductive period, post-reproductive period, total longevity, and fecundity.

During the reproductive phase, all newborn nymphs were counted and removed daily. All biological parameters were recorded individually to enable accurate statistical analysis.

Statistical analysis

The experiment was conducted under laboratory conditions following a Completely Randomized Design (CRD) with three host plants, viz., cotton, bhendi and brinjal and each treatment was replicated fifteen times. The data obtained were subjected to one-way analysis of variance (ANOVA) to determine the effect of host plants on various biological parameters of *A. gossypii.* Critical differences (5%) were calculated to compare the treatment means. Statistical analysis was performed using Web Agri Stat Package 2.0 (Jangam & Wadekar, 2004).

Results and discussion

The survival and reproductive success of insects are strongly influenced by both the type and concentration of nutrients they consume. In phytophagous insects, performance largely depends on the nutritional quality and availability of nutrients provided by their host plants.

Developmental parameters:

The developmental duration of *Aphis gossypii* varied among the three host plants (Table 1). The first instar duration differed significantly, with the shortest period recorded on cotton (1.87 ± 0.13 days), followed by brinjal (2.33 ± 0.13 days) and bhendi (2.47 ± 0.13 days). However, the durations of second, third and fourth instars did not differ significantly among hosts. The total nymphal period was significantly shorter on cotton (6.87 ± 0.22 days) compared to brinjal (8.07 ± 0.30 days) and bhendi (8.73 ± 0.25 days). The prolonged development of bhendi indicates comparatively lower suitability of this host for nymphal development. Similar results of Saha *et al.* (2016), who reported that *A. gossypii* prefers cotton plants for faster development than brinjal. Host plant characteristics such as nitrogen content, amino acid availability and secondary metabolites are known to influence aphid growth and moulting. According to Dixon (1998), host quality directly influences aphid growth rate and moulting frequency. Similarly, Blackman and Eastop (2000) reported that host plant suitability significantly affects developmental time and survival of *A. gossypii*. Faster development on cotton in the present study suggests greater nutritional suitability and efficient assimilation of plant sap. The extended developmental period observed on bhendi may reflect nutritional imbalance or the presence of defensive compounds that slow nymphal growth. Overall, cotton supported faster immature development of *A. gossypii*, indicating its suitability for rapid population build-up.

Table 1: Developmental parameters of *A. gossypii* on Cotton, Brinjal and Bhendi (days).

Host plant	Duration of instars				Total nymphal period
	I	II	III	IV	
Cotton	1.87 ± 0.13	1.80 ± 0.14	1.67 ± 0.16	1.53 ± 0.13	6.87 ± 0.22
Brinjal	2.33 ± 0.13	2.13 ± 0.17	1.87 ± 0.13	1.73 ± 0.21	8.07 ± 0.30
Bhendi	2.47 ± 0.13	2.27 ± 0.15	2.07 ± 0.12	1.93 ± 0.21	8.73 ± 0.25
CD (5%)	0.37	NS	NS	NS	0.73

All values in the table are represented as Mean ± SE; NS - Non-significant; CD - Critical difference at 5% level.

Table 2: Reproductive parameters of *A. gossypii* on Cotton, Brinjal and Bhendi.

Host plant	Pre-reproductive period (days)	Reproductive period (days)	Post-reproductive period (days)	Adult longevity (days)	Fecundity (No. of nymphs/adult)
Cotton	1.73 ± 0.15	9.67 ± 0.30	1.40 ± 0.13	12.80 ± 0.33	25.13 ± 0.50
Brinjal	2.40 ± 0.16	9.33 ± 0.25	1.60 ± 0.16	13.33 ± 0.40	23.13 ± 0.59
Bhendi	2.40 ± 0.13	9.27 ± 0.29	2.27 ± 0.12	13.47 ± 0.48	17.87 ± 0.69
CD (5%)	0.43	NS	0.39	NS	1.71

All values in the table are represented as Mean ± SE; NS - Non-significant; CD - Critical difference at 5% level.

Reproductive parameters

Reproductive traits also varied significantly among host plants (Table 2). The pre-reproductive period was shortest on cotton (1.73 ± 0.15 days) and significantly longer on brinjal and bhendi (2.40 days). A shorter pre-reproductive phase accelerates population establishment. The reproductive period and adult longevity did not differ significantly among hosts. Fecundity was significantly highest on cotton (25.13 ± 0.50 nymphs/adult), followed by brinjal (23.13 ± 0.59), and lowest on bhendi (17.87 ± 0.69). Results were in contrast with those of Uma *et al.* (2023), who found that the fecundity was significantly higher on brinjal than on cotton. In many studies, researchers have stated that nutrition on different cultivars direct the population parameters of aphids (Goundoudaki *et al.* 2003; Kaydan *et al*. 2006; Obopile and Ositile 2010). Higher fecundity on cotton indicates better allocation of assimilated nutrients toward reproduction. Similar host-dependent fecundity variation in *A. gossypii* has been documented by Satar et al. (1999), who observed significant differences in reproductive performance across crop hosts. Although brinjal (13.33 ± 0.40 days) and bhendi (13.47 ± 0.48 days) supported slightly longer adult longevity, this did not translate into increased reproductive output, suggesting that reproductive efficiency is influenced more by host suitability than lifespan alone. Dixon (1998) also noted that fecundity, rather than longevity, is a more critical determinant of aphid population growth.

Conclusion

The present study clearly demonstrates that the host plant species exert a significant influence on the developmental and reproductive biology of *Aphis gossypii*. Among the three hosts evaluated, cotton supported the most favourable biological performance, characterized by a shorter immature developmental period, reduced pre-reproductive duration, and markedly higher fecundity. Brinjal exhibited intermediate suitability, while bhendi proved comparatively less favourable, particularly with respect to total nymphal development time and reproductive output.

The combined effects of faster development and higher reproductive potential on cotton indicate its superior suitability for sustaining robust aphid populations under laboratory conditions. Accordingly, cotton can be recommended as the most appropriate host for the efficient mass rearing of *A. gossypii*. The use of cotton as a standardized rearing substrate will facilitate the production of uniform, abundant, and healthy aphid cohorts for various experimental purposes, including toxicological bioassays, host–plant interaction studies, and evaluations of natural enemies.

Acknowledgements

I sincerely thank the Department of Entomology, Annamalai University, Tamil Nadu, for providing the necessary facilities and support throughout the course of this research.

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MS Received on 03rd February, 2026
MS Accepted on 4th March, 2026

DOI: 10.55278/UMPS6564

Range Extension and Recurrent Phoresy in *Paratelenomus anu* (Hymenoptera: Scelionidae)

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Abstract

This study reports the first record of the egg parasitoid, *Paratelenomus anu* Rajmohana, Sachin & Talamus, 2019 (Hymenoptera: Scelionidae) from Bihar, India, and documents a novel tritrophic association involving *Cajanus cajan* (L.) (Fabaceae), *Megacopta cribaria* (Fabricius) (Hemiptera: Plataspidae), and *P. anu*. The recurrence of phoresy across geographically distinct populations confirms that this behavior represents a stable and adaptive host-location strategy in the species. These findings highlight the potential ecological importance of *P. anu* as a natural biological regulator of *M. cribaria* in pulse-based agroecosystems, contributing valuable insights into its distribution, behavior, and prospective role in integrated pest management.

Keywords: Biological control, Egg parasitoid, Lablab bug, Pigeon pea, Tritrophic interaction

Introduction

Tritrophic interactions among plants, herbivorous insects, and their natural enemies constitute a fundamental ecological framework influencing pest population dynamics in agroecosystems (Price *et al*., 1980; Fatouros and Huigens, 2012). Pigeon pea (*Cajanus cajan* (L.) Huth) (Fabaceae) is one of the most important pulse crops cultivated in India and supports a diverse assemblage of phytophagous insects (Sharma *et al*., 2010; Pal and Tiwari, 2020). Among them, *Megacopta cribaria* (Fabricius) (Hemiptera: Plataspidae) is a sap-feeding plataspid bug widely recognized for its polyphagy and invasive potential. The species is reported as a voracious feeder on kudzu and numerous cultivated crops including soybean (Medal *et al*., 2016), lablab bean (Schaefer and Panizzi, 2000; Rajmohana *et al*., 2019), pigeon pea (Hoffmann, 1932), *Phaseolus* spp. (Hoffmann 1931; Easton and Pun, 1997), broad beans (Ishihara, 1950), peach (*Amygdalus persica*) and jujube (*Ziziphus jujube*) (Wang *et al*., 1996; Li *et al*., 2001; Wang *et al*., 2004). Its invasive capacity (Suiter *et al*., 2010; Gardner *et al*., 2013) and wide host range underscore its economic significance in legume-based agroecosystems.

Egg parasitoids represent a major source of natural mortality in *M. cribaria*. A diverse complex of hymenopteran parasitoids has been recorded from its eggs, including *Encarsia boswelli* (Girault) (Aphelinidae), *Ablerus* spp. (Azotidae), *Ooencyrtus nezarae* Ishii (Encyrtidae), *Trissolcus latisulcus* Crawford, and *Paratelenomus saccharalis* (Dodd), *P. tetartus* (Crawford) (Scelionidae) (Eger *et al*., 2010; Rider 2018; Rajmohana *et al*., 2019). In pulse-based agroecosystems, egg parasitoids are particularly important because they intercept pest populations at the earliest developmental stage. *Paratelenomus anu* Rajmohana, Sachin & Talamus, 2019 was described from Southern India (Kerala, Karnataka and Tamil Nadu) as a phoretic egg parasitoid of *M. cribaria* on *Lablab purpureus* L. (Fabaceae) (Rajmohana *et al*., 2019; OSU-MBD database: https://mbd-db.osu.edu/).

The present study reports the range extension of *P. anu* and also explores the tritrophic association documented in Bihar (Fig. 1) India, involving *Cajanus cajan*, *M. cribaria*, and *P. anu*.

Materials and Methods

Adult specimens of *M. cribaria* were collected from pigeon pea (*Cajanus cajan*) fields at Kawar Lake Bird Sanctuary, Begusarai district, Bihar, India, on 06 December 2025 (Fig. 2a, b). Specimens were collected along with fresh host foliage and transported live to the Gangetic Plains Regional Centre, Zoological Survey of India (ZSI-GPRC) laboratory. The bugs were maintained in transparent glass containers covered with muslin cloth for aeration and supplied regularly with fresh pigeon pea twigs. Egg masses laid within the container in two batches were retained for observation (Fig. 3c, d). All emerged insects, the nymphs and the hymenopteran parasitoids were collected and preserved in absolute ethanol.

The host bugs and parasitoids were subsequently point-mounted for examination (Fig. 2b; Fig. 3a, b). Specimens were examined and photographed using Leica EZ4 E and Leica M205A stereomicroscopes, and images were captured and processed using Leica Application Suite (LAS) software.

Identification of the host bug was carried out using standard taxonomic keys for Plataspidae (Distant, 1902; Schaeffer & Panizzi, 2000). Parasitoid identification was based on the original description of *Paratelenomus anu* and relevant taxonomic literature on Scelionidae (Masner, 1972; Rajmohana *et al*., 2019; Rajmohana, 2025). Voucher specimens examined in this study have been deposited in the National Zoological Collections (NZC), Zoological Survey of India (ZSI), Kolkata.

Results

The field collected bugs oviposited on pigeon pea twigs within the rearing container (Fig. 3c, d), in the laboratory. A total of 20 hymenopteran parasitoids (17 ♀, 3 ♂) and four bug nymphs emerged in the rearing container, though no parasitoids were intentionally introduced. Parasitoid emergence was recorded on the 30th day following collection under ambient winter conditions.

Morphological examination confirmed the parasitoid as *Paratelenomus anu* Rajmohana, Sachin & Talamus, 2019 (Fig. 3a, b). Antennal segmentation and metasoma details being consistent with Telenominae, while other morphological characters like sculpture on basal frons, and nature of metasoma supported placement in *Paratelenomus* Dodd (Masner, 1972). The distinctly bidentate labrum apex, nature of submedian carina on the frons, and consistent characteristic sculpture on mesoscutum, mesopleuron and dorsum of basal tergites matched fully to the original description of *P. anu* (Rajmohana *et al*., 2019).

The host was identified as *Megacopta cribaria* (Fig. 2b) based on typical plataspid characters, body greenish yellow to light brown, beneath black, apices of lateral lobes of head completely meeting in front of central lobe, pronotum contains two discal transverse angulated striae and a sublateral series of punctures in each anterior lateral dilatation, scutellum with the basal callosity well-defined by a marginal impression, and connected on each side with the lateral margin by a continuous linear series of punctures. (Distant, 1902; Schaeffer and Panizzi, 2000).

Discussion

The present record extends the known distribution of *Paratelenomus anu* from Kerala, Karnataka and Tamil Nadu, in southern India to Bihar in the eastern Gangetic plains (Fig. 1). The record also expands the ecological interface previously known for the parasitoid, which was earlier associated with *Lablab purpureus* (Rajmohana *et al*., 2019), the novel tritrophic linkage documented here being *Cajanus cajan–Megacopta cribaria–Paratelenomus anu*.

A notable feature of the present study is the emergence of parasitoids from host eggs laid entirely under laboratory conditions (Fig. 3c, d). Only adult *Megacopta cribaria*, together with pigeon pea twigs, were introduced into the rearing container, and oviposition occurred exclusively within the enclosure. Such cases of parasitoid emergence from the host eggs can only be attributed to prior phoretic attachment of fertilized female wasps to field-collected adult bugs. The parasitoids due to their minute size could have escaped detection during field collection of the host bugs. *Paratelenomus anu* was previously documented as a phoretic egg parasitoid of *M. cribaria* (Rajmohana *et al*., 2019), and the present observation independently corroborates this biological trait. Phoresy in scelionids is recognized as an adaptive host-location strategy that facilitates precise synchronization between parasitoid oviposition and host egg deposition (Huigens and Fatouros, 2013; Fatouros *et al*., 2020). The extended emergence window as recorded in the present study is consistent with observations on scelionid egg parasitoids of stinkbugs and related heteropterans, in which cool winter temperatures and overwintering conditions greatly prolong the developmental period (Diedrick, 2023).

The recurrence of phoresy across geographically distinct populations indicates that host attachment is a stable and adaptive trait in *P. anu*. This strategy facilitates precise exploitation of freshly laid eggs of its host bug *M. cribaria*. Although field-based evaluations are necessary, the finding overall underscores the potential role of *P. anu* in the natural regulation of the bug populations in pulse-based agroecosystems.

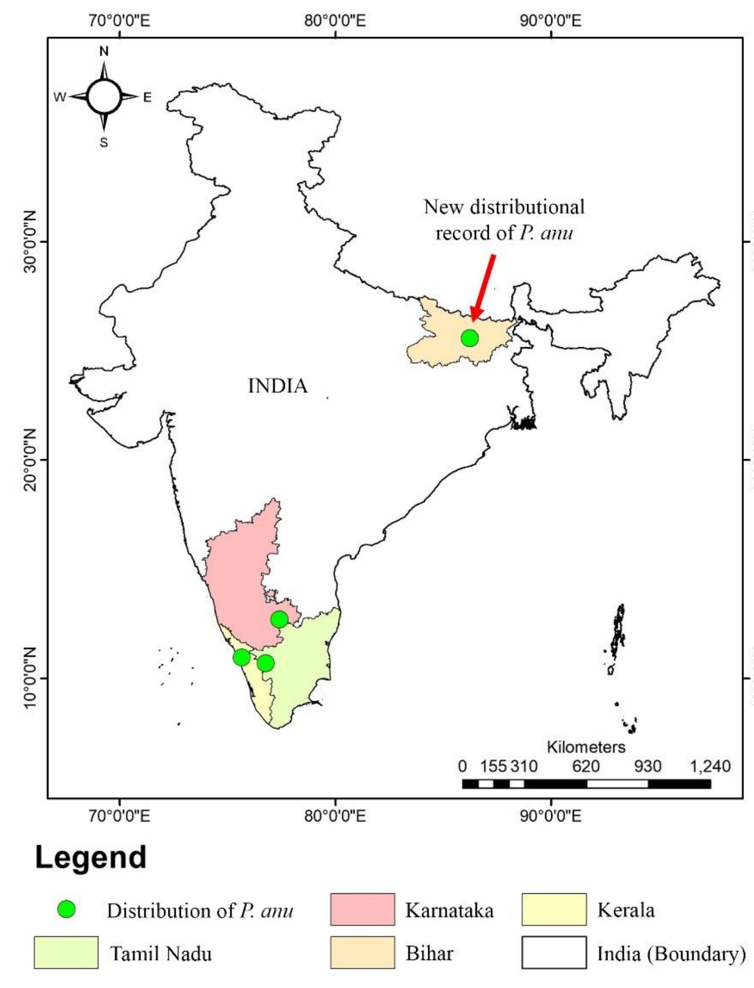

Fig. 1: Distribution of *Paratelenomus anu* Rajmohana, Sachin and Talamus in India

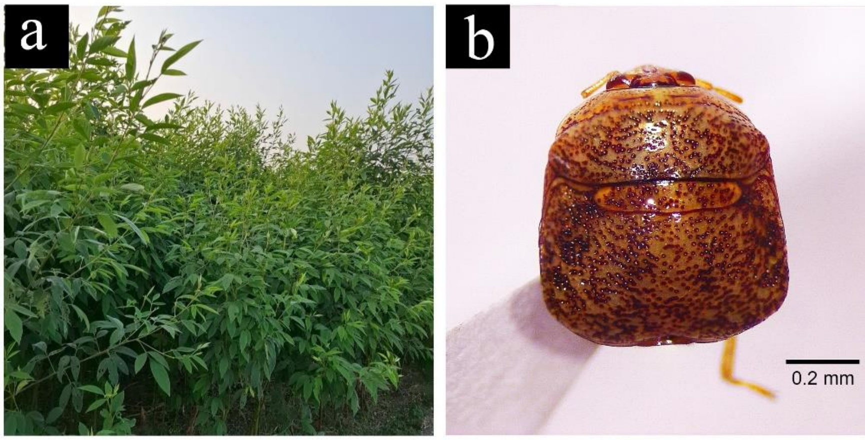

Fig. 2: a) Pigeon pea agro-ecosystem b) Adult *Megacopta cribaria* (Fabricius) ♂

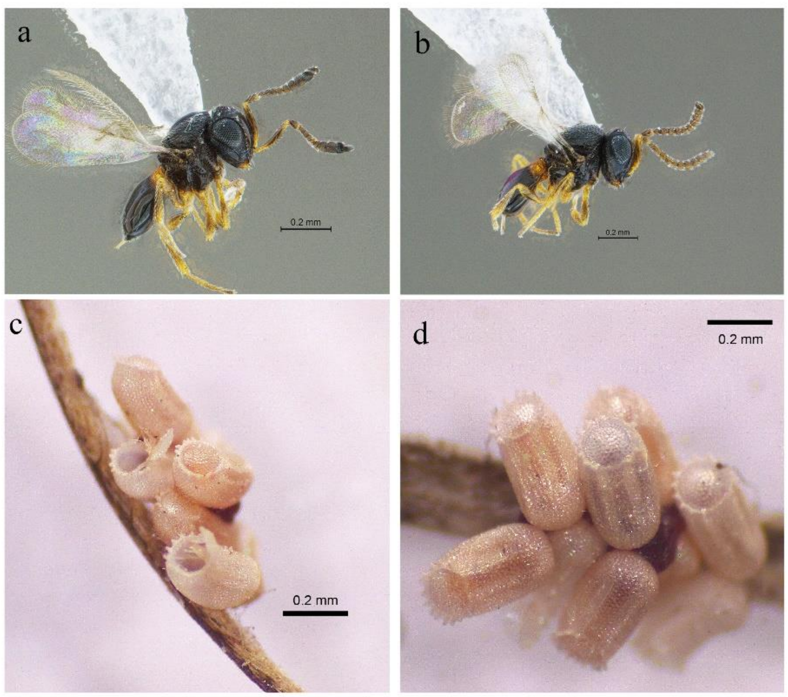

Fig. 3: a) *P. anu* (Female) b) *P. anu* (Male) c & d) Eggs of *M. cribaria*

Acknowledgements

The authors express sincere gratitude to the Director, Zoological Survey of India, Kolkata, and the officer-in-charge ZSI, GPRC, Patna for providing necessary research facilities to undertake this study. JA is grateful to the Council of Scientific and Industrial Research (CSIR) for the award of a Senior Research Fellowship [09/1181(0017)/2021-EMR-I]. SP and JA extends appreciation to their labmates for continuous support.

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MS Received on 19th January, 2026
MS Accepted on 20th February, 2026

DOI: 10.55278/IFNE2683

First Record of *Spilosoma obliqua* (Walker) (Lepidoptera: Erebidae) Infestation on the Invasive Weed *Lantana camara* L. from Uttar Pradesh, India

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Abstract

The gregarious larval stage of the Bihar hairy caterpillar, *Spilosoma obliqua* (Walker), was observed infesting the invasive weed *Lantana camara* L. at C. S. Azad University of Agriculture and Technology, Kanpur, on 1 December 2025. Larvae collected from the field were reared on *L. camara* under laboratory conditions at ICAR–IIPR, Kanpur. Successful completion of the entire life cycle—including pupation, adult emergence, and subsequent oviposition—confirmed L. camara as a suitable and reproductively supportive host for *S. obliqua*. This constitutes the first confirmed record of *S. obliqua* utilizing *L. camara*, indicating a significant host range expansion. The ability of this polyphagous pest to survive and reproduce on an invasive weed highlights its potential role in weed suppression, while simultaneously raising concerns for its movement between weed reservoirs and adjacent crops. These findings provide new insights into host–pest interactions and have implications for integrated weed and pest management strategies in Uttar Pradesh and other regions where L. camara is widespread.

Key words: *Spilosoma obliqua*, *Lantana camara*, new host record, Uttar Pradesh, Weed management

Introduction

The Bihar hairy caterpillar, *Spilosoma obliqua* (Walker) (Erebidae: Lepidoptera), is a highly polyphagous defoliator responsible for serious yield losses in a wide range of agricultural and horticultural crops across India (Sujayanand *et al*., 2019). It is known to feed on over 100 cultivated and non-cultivated plant species and frequently reaches outbreak levels in several cropping systems. Despite its broad host range and economic importance, the pest's association with non-crop vegetation—particularly invasive weeds that often serve as off-season refugia and alternative food sources—remains insufficiently understood. The role of weeds as ecological reservoirs for polyphagous pests has been widely emphasized, as they can significantly influence pest population persistence and seasonal carryover (Norris & Kogan, 2005).

Lantana camara L. (Verbenaceae), a globally invasive woody shrub, dominates large swathes of degraded landscapes in India, including the Indo-Gangetic plains and forest margins of Uttar Pradesh. The species possesses a diverse array of bioactive secondary metabolites, including lantadenes with known insecticidal and antifeedant properties that generally deter herbivory (Ghisalberti, 2000; Sharma *et al*., 2007).

The present study documents, for the first time, the successful development and infestation of *S. obliqua* on *L. camara* in Uttar Pradesh, India. This new regional record expands the known ecological amplitude of *S. obliqua*, demonstrating its capacity to utilize an invasive, chemically defended weed as a viable host. Such an association has important ecological and pest-management implications, particularly given the widespread abundance of *L. camara* in northern India, where it may function as an off-season reservoir supporting pest survival and dispersal. This finding highlights the species' remarkable ecological plasticity and underscores the need to reassess its host range dynamics in heterogeneous agro-ecological landscapes.

Materials and Methods

First-instar larvae of *Spilosoma obliqua* (approximately 65 individuals) were observed feeding on *Lantana camara* at the Vegetable Science Department, C.S.A.U.A.&T., Kanpur, Uttar Pradesh (26.4499°N, 80.3319°E). The larvae were carefully collected using sterile forceps and transferred to disinfected insect-breeding dishes. They were then transported to the Bio-ecology Laboratory at ICAR–IIPR, Kanpur for mass rearing.

Rearing was carried out under controlled environmental conditions in a BOD incubator set at 25 ± 1 °C temperature and 65 ± 5% relative humidity. Larvae were provided fresh, pesticide-free *L. camara* leaf twigs daily, ensuring continuous access to adequate foliage throughout their development.

Regular observations were recorded on larval survival, feeding behavior, developmental progression, pupation, and adult emergence. Species identity was confirmed through the examination of both larval and adult morphological characters using standard entomological taxonomic keys.

Results and Discussion

Larvae of *S. obliqua* readily accepted *L. camara* foliage under laboratory conditions and successfully completed their development, culminating in normal pupation and adult emergence. Field observations of an aggregation of approximately 60–65 first-instar larvae exhibiting characteristic gregarious feeding behavior further confirmed the early instar stage and validated *L. camara* as a suitable substrate for oviposition and early larval survival.

Although *S. obliqua* is a highly polyphagous species, its association with *L. camara* has not previously been reported from Uttar Pradesh. In contrast, its occurrence on *L. camara* was earlier documented in Madhya Pradesh by Mishra et al. (2013). Additionally, Tripathi et al. (2017) demonstrated the potential of *S. obliqua* as a biological control agent against *L. camara*, comparing its defoliation efficacy with that of *Rhizopus* species. The present findings further substantiate the species' ability to complete its life cycle on this invasive weed.

These results align with the observations of Palmer and Pullen (1995), who reported seven different species of Arctiidae defoliators as phytophagous insect pests in tropical America. Successful larval development on *L. camara*—a plant known to contain insecticidal and antifeedant secondary metabolites such as the triterpenoids Lantadene A and Lantadene B, and flavonoids including hispidulin, pectolinarigenin, and pectolinarin (Ghisalberti, 2000; Sharma et al., 2007)—indicates the physiological adaptability of *S. obliqua*. This suggests that the species possesses an effective detoxification system typical of highly polyphagous lepidopterans, allowing it to tolerate or metabolize bioactive compounds present in the host foliage.

From an ecological perspective, *L. camara* may serve as an alternative or refuge host that facilitates the persistence of *S. obliqua* populations during crop-free periods. Such off-season reservoirs are known to influence pest carryover and may contribute to subsequent outbreak dynamics in adjoining agro-ecosystems (Norris and Kogan, 2005). The ability of *S. obliqua* to utilize this widespread invasive weed highlights the importance of considering non-crop vegetation when developing integrated pest management strategies.

Conclusion

This study reports the first confirmed record of *S. obliqua* infestation on *L. camara* in Uttar Pradesh, India. The species was not only found naturally occurring on the weed but was also able to complete its entire life cycle under laboratory conditions, thereby establishing *L. camara* as a true host. Consistent feeding across larval instars on tender shoots as well as mature leaves demonstrates that the weed provides adequate nutritional and physiological support for development. These findings broaden the known host range of *S. obliqua* and emphasize the need to monitor egg masses and early larval aggregations on *L. camara*, particularly in landscapes where the weed is abundant. Targeting infestations on *L. camara* may offer an effective strategy to suppress early-season populations and reduce subsequent pest pressure in agricultural systems.

Fig. 1. (A, B) *L. camara* leaves infested by BHC. **(C)** Third-instar larva feeding on a leaf twig. **(D)** Fourth-instar larva defoliating tender shoots and young leaves. **(E)** Larva feeding on mature leaf. **(F)** Chlorophyll scraping on leaves by 3rd instar. **(G)** Adult male and female of BHC emerged from rearing on *L. camara*. **(H)** Egg masses of BHC.

Acknowledgements

The author sincerely acknowledges the Director, ICAR-Indian Institute of Pulses Research (IIPR), Kanpur, for providing laboratory facilities to carry out the present study

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MS Received on 23rd January, 2026
MS Accepted on 28th February, 2026

Review articles & Short notes

DOI: 10.55278/FYNH6543

Evolution, Composition and Functional Mechanisms of Lepidopteran Venoms: A Comprehensive Review

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Abstract

Animal venoms are of considerable scientific interest due to the risks they pose and their value in understanding evolution, ecology, pharmacology, biodiscovery and biotechnology. In Lepidoptera, venom systems have evolved independently multiple times, primarily as defensive adaptations that protect the larval stages from predators. Although these venoms are consistently produced by cuticle-derived structures and specialized setae, they display remarkable variation in chemical composition, mode of action and biological effects, highlighting their diverse evolutionary origins. Lepidopteran envenomation most commonly causes pain, irritation and inflammatory responses in humans; however, certain species can induce severe or fatal outcomes, including hemorrhagic syndromes in humans and veterinary conditions such as equine amnionitis and fetal loss. Despite their medical and ecological importance, only a small number of venom components have been fully characterized to date. Key examples include coagulation-disrupting toxins from *Lonomia obliqua* (Saturniidae) and nociceptive cecropin-like peptides from *Doratifera vulnerans* (Limacodidae). This review synthesizes current knowledge on the evolution, diversity and mechanistic functions of lepidopteran venoms and highlights significant gaps that warrant future investigation.

Keywords: Lepidoptera, venom, chemical diversity, envenomation, peptide, evolution

Introduction

Venoms is a specialized toxic secretion produced by an animal, stored in a venom-gland or equivalent apparatus and actively delivered into another organism typically *via* bite, sting or other wound to immobilize, kill or deter that organism (Jenner *et al*., 2025). Venoms are considered rich natural repositories of diverse molecules, much like the synthetic chemical libraries commonly screened by medicinal chemists and biotechnologists for specific biological activities. As such, venoms are widely acknowledged as important sources of novel compounds with significant potential applications in science, medicine and biotechnology (Reynaud *et al*., 2020). Lepidoptera is the second most diverse order of insects, comprising approximately 1,80,000 species. Among these, venom production and utilization have been documented in 576 species across 14 different families. Overall, about 3,260 lepidopteran species are considered potentially venomous, representing only around 2 per cent of the entire order (Battisti *et al*., 2024). The actual number of venomous species is likely considerably higher, as data are lacking for many species. For instance, a recent study on *Lonomia* spp. revealed that although envenomations have traditionally been attributed to *Lonomia obliqua* or *Lonomia achelous* the genus actually comprises around 60 species (Walker, 2024).

Evolution

Venom production in Lepidoptera has evolved independently multiple times across several families, demonstrating striking convergent evolution with other venomous lineages such as Hymenoptera and Arachnida. Despite their independent origins, some venom components exhibit notable functional parallels with toxins found in distantly related taxa. For example, structurally similar inhibitor cystine knot (ICK) peptides have been identified in phylogenetically distant species such as *Acharia stimulea* and *Parasa lepida* (Mitpuangchon *et al*., 2021).

This repeated evolution of venom systems within Lepidoptera highlights strong selective pressures on larval defense, particularly against predators and parasitoids. The diversity in venom chemistry and deployment strategies reflects the ecological niches occupied by different lepidopteran families and underscores the importance of larval defensive evolution in shaping venom complexity.

Venom production and delivery

Battisti *et al.* (2011) categorized lepidopteran venom-delivery structures into true setae, modified setae and spines, each representing distinct morphological and functional adaptations.

- True setae are small, hollow, detachable structures (50–200 µm) filled with toxins secreted by trichogen cells. Upon physical contact or disturbance, these setae readily break off and penetrate the skin, releasing venom.
- Modified setae possess an additional gland cell that secretes venom into an internal reservoir, allowing more controlled or prolonged delivery.
- Spines, which are larger epidermal projections, contain glandular cells that release their toxic contents when the spine tip is broken or compressed.

These structures are highly effective defensive mechanisms, particularly in larval stages. Murphy *et al.* (2010) demonstrated that limacodid caterpillars armed with dense stinging spines exhibit significantly higher survival rates against invertebrate predators compared to unspined or lightly spined larvae. This provides strong evidence that venom and its delivery systems confer substantial adaptive advantage, shaping the evolution of larval defensive morphology across venomous Lepidoptera.

Composition and function of venoms

Venoms from various lepidopteran families display significant biochemical diversity and distinct physiological effects. Saturniidae venoms, particularly from *Lonomia* spp. contain serine proteases, phospholipase A_2 and procoagulant factors that induce hemorrhage and coagulopathy (Veiga *et al.,* 2005). Limacodidae venoms from *Doratifera vulnerans* and *Acharia stimulea* are rich in cecropin-like and ICK peptides causing intense pain and inflammation. Megalopygidae venoms from *Megalopyge opercularis* include pore-forming megalysins and CRiSPs that trigger cell lysis and strong inflammatory responses (Walker, 2024). Whereas *Megalopyge lanata* and *Podalia orsilochus* extracts cause tissue damage ranging from mild inflammation and leukocyte infiltration to severe necrosis and haemorrhage (Sanchez *et al.*, 2019).

Notodontidae venoms, mainly from *Thaumetopoea pityocampa* and *Ochrogaster lunifer* species, contain allergenic proteins triggering severe skin and respiratory inflammation (Cawdell-Smith *et al.,* 2012). Exposure of shaved mice to *T. pityocampa* caterpillars causes degranulation of mast cells (Kalender *et al.,* 2004). Erebidae venoms, for example the venom of *Premolis semirufa contain* histamine, kinins and proteases responsible for dermatitis and chronic inflammatory conditions (Siqueira *et al*., 2021). Similarly, lasiocampidae venoms from *Dendrolimus punctatus* and *Malacosoma americanum* cause systemic inflammation and reproductive disorders, reflecting the multifunctional toxicity of lepidopteran venoms (Sebastian *et al.,* 2008).

Molecular mode of action

i) Lopap

Lopap is a member of the lipocalin protein family that exhibits prothrombin-activating activity. Lopap selectively activated prothrombin, but not Factor X and could not clot fibrinogen without prothrombin. It functions as a calcium-dependent serine protease, generating prothrombin degradation products similar to those produced by Factor Xa, including active prothrombin-2 and α-thrombin. Reis *et al.* (2001) reported that administering purified Lopap to rats led to thrombus formation in small blood vessels, rendered the blood incapable of clotting and caused depletion of fibrinogen and platelets, while leukocyte and erythrocyte levels remained unaffected.

ii) Losac

Losac is a member of the hemolin protein family that exhibits Factor X–activating activity. Losac was shown to enhance human endothelial cell proliferation, elevate nitric oxide levels and stimulate the release of tissue plasminogen activator (Flores *et al.*, 2006). Recombinant Losac (rLosac) expressed in *Escherichia coli* shortened blood clotting time in a dose-dependent manner without directly degrading fibrin or fibrinogen. Its activity depended on the presence of Factor X, as it was ineffective in Factor X-deficient blood but remained active in blood lacking other coagulation factors (Flores *et al*., 2011).

iii) Megalysins

Megalysins from the puss caterpillar cause severe tissue damage by forming membrane pores that destroy cells and basement membranes (Walker *et al.*, 2023).

iv) Limacodid Venom Toxins

The peptides Δ-LCTX$_2$-Dv11 and Δ-LCTX$_2$-Dv12 from *D. vulnerans* venom are pain-inducing, cationic and amphipathic molecules that lack disulfide bonds and share structural similarities with hymenopteran aculeatoxins. Dv11 and Dv12 exhibit membrane-permeabilizing activity comparable to cecropins and aculeatoxins like melittin. Unlike other cecropin family peptides, they lack a conserved proline residue and C-terminal amidation, features that may be linked to their specialized role as venom toxins (Walker *et al.*, 2021). These peptides are members of the cecropin family, originally identified in the pupae of *H. cecropia* and are known for their antimicrobial properties (Steiner *et al*., 1981).

Use as pesticides

Walker *et al.* (2021) demonstrated that several cecropin-like venom peptides-Δ-LCTX2-Dv11, Δ-LCTX2-Dv12 and U-LCTX7-Dv63—exhibit potent membrane-disruptive insecticidal activity against *Drosophila melanogaster*. Additionally, recombinant U-LCTX3-Dv33 showed strong anthelmintic effects, inhibiting the development of the parasitic nematode *Haemonchus contortus* at low micromolar concentrations. In a later study, Walker *et al.* (2023) reported that venoms from *Megalopyge opercularis* and *M. crispata* displayed time-dependent toxicity with decreasing LD_{50} values against *D. melanogaster*, indicating progressive potency or cumulative effects upon prolonged exposure.

Goudarzi *et al.* (2023) further showed that synthetic RF-amide peptides derived from *Acharia stimulea* U-LCTX13-As2, U-LCTX13-As11 and U-LCTX13-As54 caused marked insecticidal activity when injected into *D. melanogaster*. Beyond insect targets, *A. stimulea* venom was found to significantly disrupt larval development of *H.*

contortus, highlighting its potential application in veterinary parasitology.

Collectively, these findings reveal that lepidopteran venoms harbor broad-spectrum bioactivities, including:

- Insecticidal membrane-disruption
- Neuroactive and nociceptive pathways targeting invertebrate physiology
- Anthelmintic activity relevant to livestock health
- Potential synergy with existing biocontrol agents

Potential Applications in Biopesticide Development

The growing interest in lepidopteran venom components stems from their:

1. High specificity toward insect and invertebrate targets, reducing risk to vertebrates.
2. Rapid mode-of-action, often through membrane lysis or neuromodulation.
3. Structural stability, especially in ICK peptides known for resistance to heat and proteolytic degradation.
4. Evolutionary refinement, as many toxins have been sculpted under strong natural selection for deterring predators.

Several lepidopteran venom peptides share mechanistic similarities with toxins used in modern biopesticides, such as:

- Spider-derived ω-conotoxins used in recombinant baculovirus bioinsecticides
- Scorpion β-toxins engineered into entomopathogenic fungi
- Bee and wasp peptides informing synthetic insecticidal peptide design

Given this convergence, lepidopteran venoms represent a largely untapped resource for the development of next-generation biological pesticides.

Future Directions

To translate lepidopteran venom peptides into viable pest-management tools, future research should prioritize:

- Molecular characterization of unstudied venom components
- Recombinant expression systems to enable scalable production
- Toxicity screening against major agricultural pests (e.g., lepidopteran, hemipteran and coleopteran species)
- Formulation strategies for stability and field application
- Off-target and environmental safety assessments

As genomic and proteomic techniques continue to advance, many more bioactive venom peptides are likely to be discovered, greatly expanding the potential of lepidopteran venoms as eco-friendly biopesticides.

Conclusion

Venoms have evolved multiple times in lepidoptera, mostly as defensive adaptations that protect the larval life stages. While venoms are always produced in structures derived from cuticle and setae, they are diverse in their composition and bioactivity, reflecting their multiple evolutionary origins. The most common result of envenomation by lepidopterans is pain and inflammation, but envenomation by some species causes fatal hemorrhagic syndromes or chronic inflammatory conditions in humans or veterinary pathologies such as equine amnionitis and fetal loss. The handful of lepidopteran venom toxins that have been characterized includes coagulotoxins from *Lonomia obliqua* and pain-causing cecropin-like peptides from *Doratifera vulnerans*. For example, venoms from *Doratifera vulnerans* and *Acharia stimulea* exhibit strong insecticidal and nematicidal activities, effectively paralyzing insects and inhibiting nematode larval development.

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MS Received on 03rd February, 2026
MS Accepted on 03rd March, 2026

DOI: 10.55278/UKAO6166

Evaluating Tobacco (*Nicotiana tabacum*) as a Potential Trap Crop for Managing *Thrips parvispinus* in Chilli

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Thrips parvispinus is native to the Asian tropics and has been reported from Indonesia, India, Thailand, Malaysia, Singapore, Taiwan, China, the Philippines, Australia, and the Solomon Islands (Mound and Collins 2000). In India, it was later recorded on papaya (*Carica papaya*) (Tyagi et al. 2015), *Dahlia rosea* (Rachana et al. 2018), and chilli (*Capsicum annuum*) from Andhra Pradesh (Sireesha et al. 2021). Since then, the pest has been observed on hundreds of cultivated crops and weed species and continues to expand its host range. It is commonly known as Taiwanese thrips, Southeast Asian thrips, or tobacco thrips.

The pest was first observed in chilli in the Chilakaluripeta and Pratipadu mandals of Guntur district (16.09 N, 80.16 E and 16.16 N, 80.22 E) during January 2021; subsequently, its spread was noted across all chilli-growing areas of Andhra Pradesh. Later in 2021, systematic surveys were undertaken in the major chilli-growing districts of Andhra Pradesh, viz., Guntur, Krishna, Prakasam, Palnadu, and NTR. During the survey in Prakasam district, specific observations were made in tobacco fields because the pest is commonly known as tobacco thrips.

In Andhra Pradesh, Flue-Cured Virginia (FCV) tobacco is grown on approximately 125,000 ha in the East Godavari, West Godavari, Khammam, Krishna, Guntur, Prakasam, Nellore, Karimnagar, and Warangal districts, with a total production of about 170 million kg of leaf. Tobacco is an important cash crop in Prakasam district and supports livelihoods and income generation (Padmaja et al. 2015). In Prakasam district, the crop is cultivated over ~65,000–73,601 ha. Hence, while documenting *T. parvispinus* on chilli, we also recorded its incidence in tobacco fields. Subsequently, a few tobacco plants were collected from a nursery and planted as a border crop in an experimental chilli field at the Horticultural Research Station, Lam, Guntur **(Fig. 1)**.

During the survey, the maximum thrips population—particularly adults—was predominantly observed in tobacco flowers. In the experimental field, tobacco plants grown between chilli rows showed that many adult thrips were immobilized and their movement was restricted by leaf trichomes, whereas adults present in flowers remained active **(Fig. 2)**. Trichomes are natural hair-like structures on aerial plant surfaces involved in defence. They may be glandular or non-glandular and can secrete exudates that trap insects, slow their movement, and act as physical barriers—either preventing attack or reducing survival and population growth (Patel 2024). On tobacco, thrips present on leaves were often killed after becoming entrapped in trichomes; sticky leaf exudates also aided in trapping adults.

Based on these observations, we tested crude extracts prepared from dried and fresh tobacco leaves on chilli; however, these treatments did not produce convincing control of thrips in chilli. Overall, the findings indicate that tobacco can be effectively used as a trap or border crop under a push–pull strategy. Although tobacco is a preferred host for thrips, the pest is unable to complete its life cycle on tobacco because of strong inherent defense mechanisms. Moreover, populations of *Nesidiocoris tenuis*—a generalist predator—were consistently observed on tobacco, with all developmental stages active on the plants. In contrast, *N. tenuis* population was less on chilli compared to tobacco. Therefore, tobacco offers a dual advantage for managing *T. parvispinus*:

1. It attracts and kills *T. parvispinus* via natural plant defenses.
2. It serves as a reservoir for the predator *Nesidiocoris tenuis* **(Fig. 3).**

Fig. 1: Field View of Chilli Experimental plot where Tobacco was planted between rows

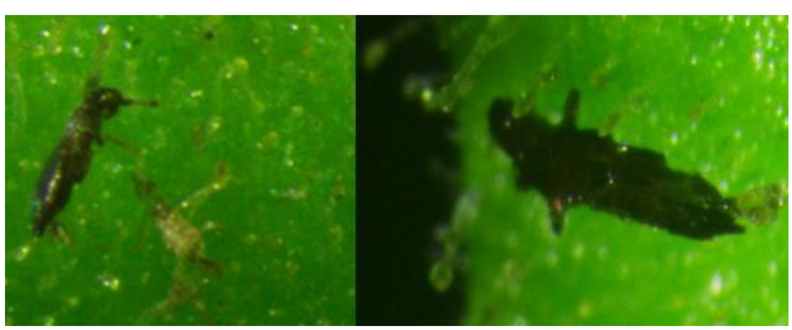

Fig. 2: Microscopic observation showing immobilised *Thrips parvinus* on Tobacco leaf

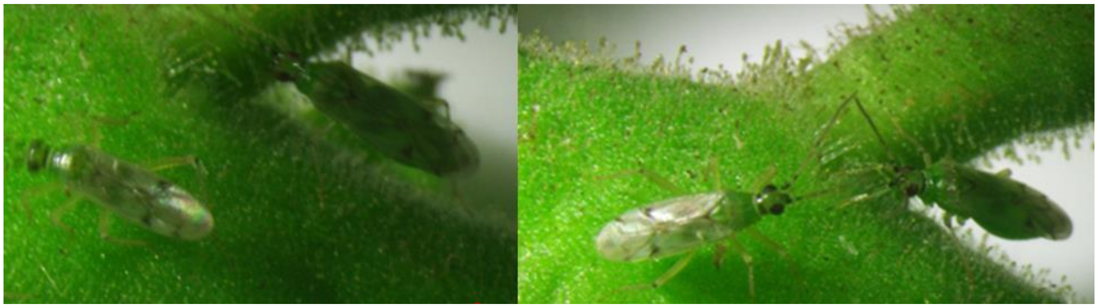

Fig. 3: *Nesidiocoris tenuis* a generalist predator activity on Tobacco

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MS Received on 14th January, 2026
MS Accepted on 14th February, 2026

DOI: 10.55278/OPKL8214

Occurrence of Midrib Folder, *Banisia myrsusalis elearalis* Walker (Lepidoptera: Thyrididae) on Sapota in Middle Gujarat

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Abstract

A study was conducted to identify the lepidopteran pest infesting sapota in Middle Gujarat. Orchard surveys carried out during November and December, 2025 revealed infestations on sapota trees at the Horticulture Farm, College of Agriculture, AAU, Vaso and the Horticulture Farm, College of Horticulture, AAU, Anand. The percentage of leaf damage ranged from 20-24% at the Horticulture Farm, College of Agriculture, AAU, Vaso and 9-11% at the Horticulture Farm, College of Horticulture, AAU, Anand. Larvae were collected from the infested trees and brought to the laboratory of the Department of Entomology, College of Agriculture, AAU, Vaso for identification and further studies. Based on detailed morphological examination, the pest was identified as the midrib folder, *Banisia myrsusalis elearalis* Walker. This study represents the report of occurrence this lepidopteran pest infesting sapota in Middle Gujarat.

Key words: Sapota, lepidopteran pest, *Banisia myrsusalis elearalis*, midrib folder

Introduction

Sapota, *Manilkara achras* (Miller) Forsberg (syn. *Achras zapota*) is an important tropical fruit crop belonging to the family Sapotaceae. Commonly known as sapodilla or chiku in India, the fruit is native to Mexico and Central America but is now extensively cultivated across the tropics for its sweet, nutritionally rich fruits. Sapota contains 12–14 per cent sugars and a 100 g edible portion consists of 73.7 g moisture, 21.49 g carbohydrates, 0.7 g protein, 1.1 g fat and notable quantities of calcium (28 mg), phosphorus (27 mg), iron (2 mg) and ascorbic acid (6 mg) (Bose and Mitra, 1990).

The cultivation of sapota in India primarily focuses on its fruit value. India ranks first in sapota production in the world with cultivation area of 70000 ha with an annual production of 858000 MT (Anonymous, 2025a). In Gujarat specifically, sapota is growing on approximately 24622 ha, resulting in annual production of 243612 MT with an average productivity of 9.89 MT/ha. In Anand district, sapota is growing on approximately 390 ha, resulting in annual production of 3687

MT with an average productivity of 9.45 MT/ha. (Anonymous, 2025b).

Sapota is attacked by nearly 25 insect-pests in India (Butani, 1979). Among these, bud borer (*Anarsia achrasella*), midrib folder (*Banisia myrsusalis elearalis*), chiku moth (*Nephopteryx eugraphella*), leaf miner (*Achrocercops gemoniella*) and fruit flies (*Bactrocera dorsalis* Hendel, *B. zonata* Saunders) are considered major pests. Bud borers, leaf miners and midrib folders remain active throughout the year, causing varying levels of damage. Other pests such as Idioscopus spp., mealybugs (*Pseudococcus lilacinus* Cockerell), blossom thrips (*Frankliniella dampfi* Priesner), fruit flies and the weevil (*Myllocerus maculosus* Desbrochers) are regarded as minor pests. Additionally, a relatively newer pest, the sapota seed borer (*Trymalitis margarias* Meyrick) has also been reported from parts of India (Puri & Mote, 2003; Shukla, 2009).

Among the foliage-feeding insect-pests, the sapota midrib folder, *B. myrsusalis elearalis* (Lepidoptera: Thyrididae) is of increasing concern. It was first recorded in Punjab (Sandhu and Sran, 1980) followed by reports from South Gujarat on Khirnee (*Manilkara hexandra*) an important rootstock in nurseries (Jhala *et al*., 1988). Later, it was reported as a serious pest in the hill zone of Karnataka at the Zonal Horticultural Research Station (ZHRS), Mudigere (Ravulapenta *et al*., 2013). More recently, detailed re-examination of specimens previously identified as *B. myrsusalis* resulted in their reclassification as *Banisia argutula* based on morphological and genitalic characters (Martinez *et al*., 2017) highlighting the taxonomic complexity within this group.

Materials and Methods

Field surveys were conducted during November and December 2025 to record the occurrence of lepidopteran pests in sapota orchards located at the Horticulture Farm, College of Agriculture, Anand Agricultural University (AAU), Vaso and the Horticulture Farm, College of Horticulture, AAU, Anand, Gujarat. The surveyed orchards were 7–8 years old and exhibited distinct foliar damage symptoms. Random sampling was carried out to assess the extent of infestation and to document the presence of midrib folder larvae. Damage caused by the pest was characterized by terminal shoots bearing dried and folded leaves. Upon critical observation, the larvae differed morphologically from known lepidopteran pests of sapota.

Up on critical observation, larvae were differed from known lepidopteran species, they were collected and brought to the laboratory of the Department of Entomology, College of Agriculture, Anand Agricultural University, Vaso for detailed examination.

Collected larvae were reared under laboratory conditions until completion of their life cycle and all developmental stages egg, larva, pupa and adult were obtained. After adult emergence, specimens were euthanized, properly pinned and photographed using a stereo zoom microscope. Preserved specimens, along with photographic documentation were forwarded to taxonomic experts for accurate species identification.

To estimate percent leaf damage, five uniformly sized sapota trees were randomly selected in each orchard. From each tree, eight twigs (20 cm length) two from each cardinal direction were tagged and observed. The number of healthy and damaged leaves on each tagged twig was recorded at fortnightly intervals and percent leaf damage was calculated accordingly.

Results and Discussion

Field assessment conducted during November–December 2025 revealed varying levels of sapota leaf damage at the two surveyed locations. At the Horticulture Farm, College of Agriculture, AAU, Vaso, leaf damage ranged between 20–24%, whereas comparatively lower damage (9–11%) was recorded at the Horticulture Farm, College of Horticulture, AAU, Anand. These observations confirm the presence and active feeding of the midrib folder on sapota foliage in Middle Gujarat.

Sr. No.	Crop and Variety	Location	Leaf damage (%)
1	Sapota *var. Kalipatti*	Horticulture Farm, CoA, AAU, Vaso	20-24
2	Sapota *var. Kalipatti*	Horticulture Farm, CoH, AAU,Anand	09-11

Marks of Identification and Life cycle

Adult females lay eggs singly on tender leaves. Freshly laid eggs are oval with one end broader and appear dark yellow with reddish transverse bands **[Fig. 1(A)]**. A female lays about 40–42 eggs, which hatch within 3–6 days. The larva passes through four instars with early instars having a reddish-brown head capsule and brown prothoracic shield. Newly emerged larvae scrape the leaf surface and remain active for 15–18 days.

The fourth instar larva is pale green with a reddish-brown head and light brown prothoracic shield **[Fig. 1(B)]**. Mature larvae exit the folded leaves and pupate either within webbed leaves or inside the leaf fold. The pupal stage lasts 9–12 days and the freshly formed obtect pupa is reddish-brown **[Fig. 1(C)].**

Adults show sexual dimorphism:

- Males possess a dark almond-colored head, thorax and abdomen.
- Females exhibit lighter brownish coloration [Fig. 1(D)]. A distinct white spot is visible on the forewing in some individuals. The entire life cycle is completed within 34-42 days under field conditions.

Damage symptoms

The caterpillar induces folding of tender leaves and feeds internally, predominantly during the new foliage flush stage. In the early instar, the minute larvae nibble primarily along the midrib of young leaves. From the second instar onward, the larvae fold the leaf along the midrib and web it from the margins, producing a characteristic "pea-pod" appearance or they may bind three to four leaves together and feed within the enclosed structure. The larvae remain concealed inside the folded leaves and consume the green tissues. Upon maturity, they create a relatively larger exit hole in the leaf fold to emerge for pupation [Fig. 2(B)]. Infestation is evident from dried terminal shoots bearing desiccated leaves [Fig. 2(C)]. Damage has also been observed on leaves of *Khirnee* rootstocks in nursery conditions. Severe infestation of the midrib folder in newly established orchards results in stunted growth of young grafts and planted seedlings, thereby adversely affecting the crop's developmental phase.

The present investigation confirms the occurrence of the sapota midrib folder, *Banisia myrsusalis elearalis* in Middle Gujarat with measurable leaf damage across two locations. The documentation of its distinctive life cycle stages and characteristic injury symptoms provides essential baseline data for further monitoring. Considering its ability to remain active year-round and cause substantial foliage loss, timely identification and localized management strategies are critical for preventing growth suppression in young sapota orchards. This report serves as a valuable reference for future pest surveillance and integrated pest-management planning in the region.

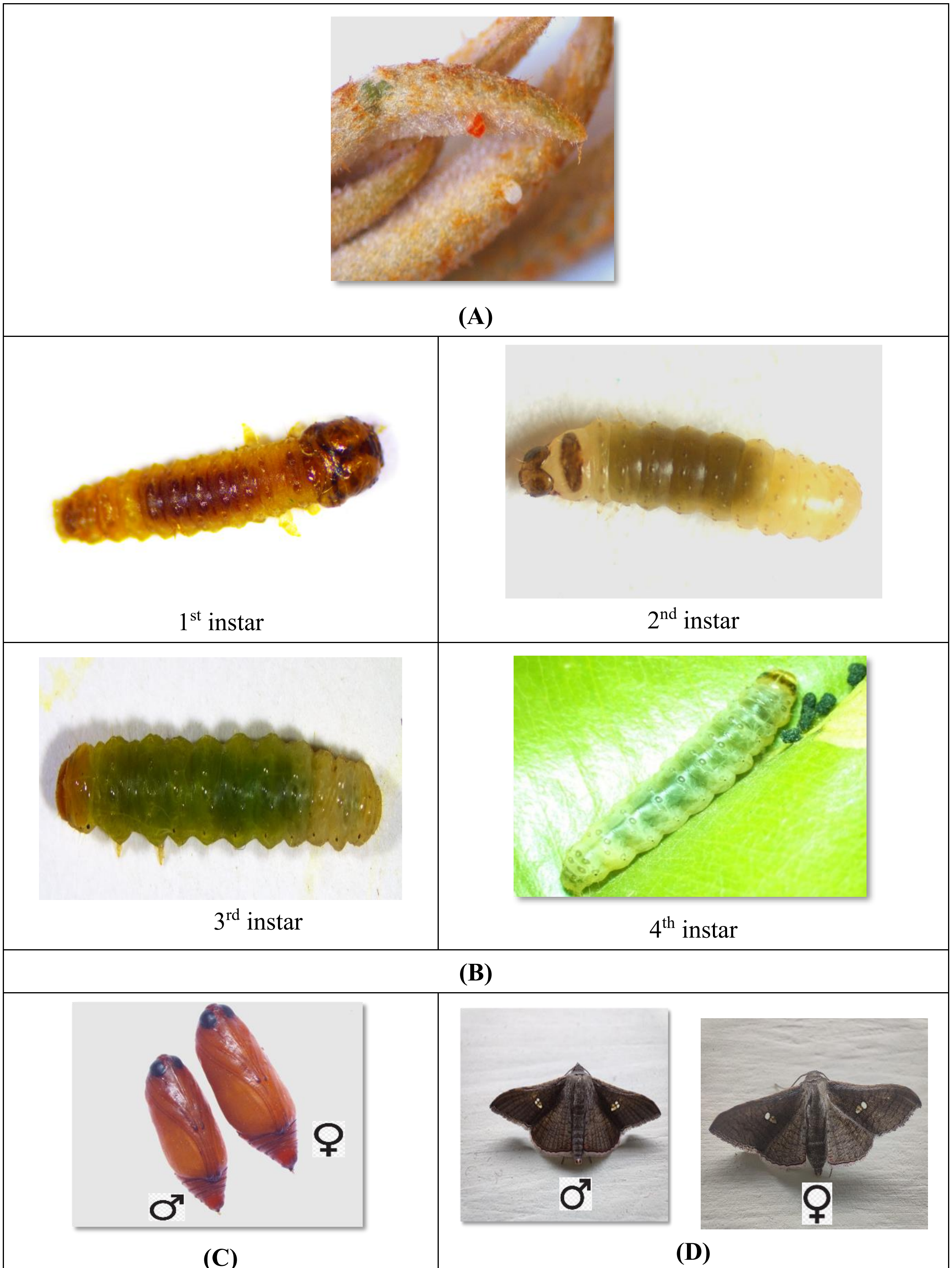
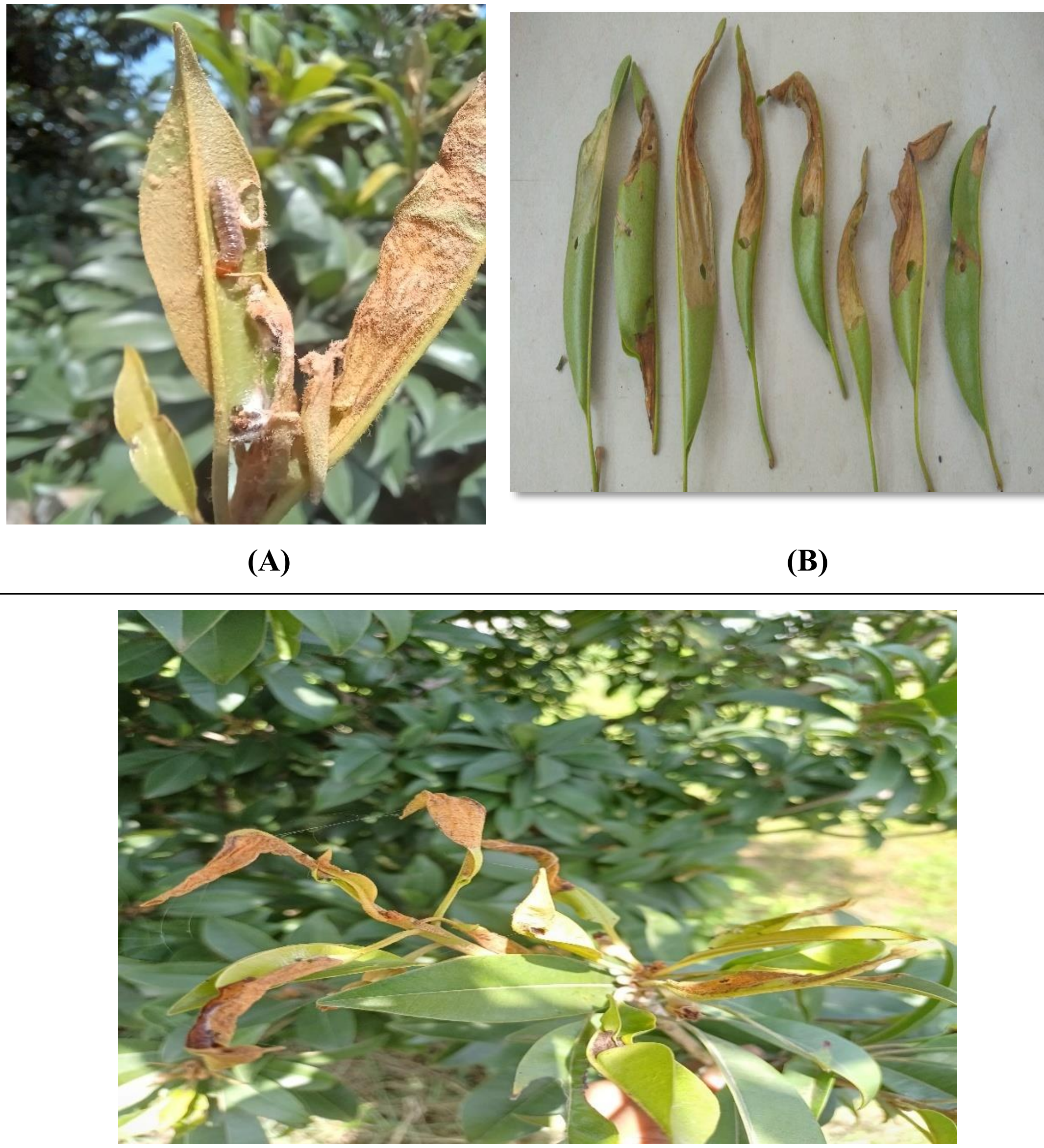


Fig. 1: (A) Egg laid on tender leaves in singly, **(B)** First, second, third and fourth instar larva, **(C)** Male and Female pupae, **(D)** Male and Female Adults



(A) (B) (C)

Fig. 2: (A) Fourth instar larva on leaves (B) "Pea-pod" shape leaves with larger exit hole (C) Dried terminal shoots with desiccated leaves

Acknowledgments

We are thankful to Dr. P. R. Shashank, Senior Scientist, Division of Entomolgy, ICAR-IARI, New Delhi and Dr. Sachin A. Gurule, Assistant Professor & Research Guide (Savitribai Phule Pune University, Pune), Dept. of Zoology & Research Center, K.T.H.M. College, Nashik for identifying the Midrib folder, *Banisia myrsusalis elearalis* Walker.

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MS Received on 1st February, 2026
MS Accepted on 1st March, 2026

INSECT LENS

Flower Mantid, Creobroter gemmatus (Hymenopodidae: Mantodea)

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Porter Wasp, Phimenes sp. (Vespidae: Hymenoptera)

Author: *Dr. Nagaraj, D.N., Project Head (Entomologist) Ento. Proteins Pvt. Ltd., Mangalore*
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Blowfly (blowing bubble)

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Flatid (unidentified)

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Unidentified Beetle
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A pair of mating stick insects
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Unidentified fly
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Assassin Bug, Cydnocoris sp. (Reduviidae: Hemiptera)

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Flesh flies, Sarcophaga sp. (Sarcophagidae: Diptera)

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Indian Common Clubtail, Ictinogomphus rapax (Gomphidae: Odonata)

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Lantana bug, Orthezia insignis (Ortheziidae: Hemiptera)

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Spider preying on House Fly, Musca domestica (Muscidae: Diptera)

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Important IE Blogs

Insect Environment Blog

Red Palm Weevil in 2026: Alerting a Global Emergency for Palm Ecosystems

25 January 2026

J R Faleiro *Ex-Indian Council of Agricultural Research (ICAR),* ***Hamadttu ElShafie,*** *International Center for Agricultural Research in the Dry Areas (ICARDA) and* ***Mohamed Ali Bob,*** *International Center for Biosaline Agriculture (ICBA-UAE)*

The red palm weevil (RPW) (*Rhynchophorus ferrugineus*), once a localized pest of coconut in South Asia, has transformed into one of the most destructive and invasive insects. Its ability to remain concealed deep within palm tissues, coupled with rapid reproduction and long-distance spread through infested planting material, has made it a formidable threat to palm biodiversity including global date palms.

Damaged date palms PC: Authors

Over the past few decades, RPW has expanded aggressively across the Middle East, Africa, Europe, the Mediterranean basin and Latin America. Today, it affects nearly 50% of date palm–growing countries and 15% of coconut-growing regions, with *Phoenix dactylifera* and *Phoenix canariensis*

Adult emerging PC: Authors

among its most severely impacted hosts. The Maghreb region (in North Africa) responsible for 15% of global date production remained free of RPW until 2008, but detections in Morocco, Libya, and Tunisia have since highlighted the pest's relentless spread. By 2026, apical infestations in date palm have become increasingly common, posing a major challenge even to well-established management systems. RPW is becoming severe in date palm plantations across India.

Management lies in early detection of infested palms. By the time external symptoms appear, internal tissues are often extensively damaged, leading to palm mortality. Early-stage infestations can respond to insecticidal intervention, but success depends on vigilant monitoring and rapid action.

Integrated Pest Management (IPM) remains the most effective strategy. Besides early detection, pheromone traps, preventive and curative chemical interventions, field and palm sanitation, removal of severely infested palms, and strict quarantine protocols are essential. However, the current situation demands stronger measures:

- Enhanced surveillance technologies such as acoustic sensors and thermal imaging.
- Biological control exploration, using natural enemies.
- Breeding and deploying RPW-resistant palm varieties, through CRISPR based solutions.
- Zero-tolerance policies on movement of uncertified planting material.

Grub of red palm weevil PC: Authors

In 2026, the RPW crisis is not merely an agricultural problem but is an ecological and economic emergency. Protecting palm landscapes requires coordinated international action, scientific innovation, and unwavering vigilance. The future of palm ecosystems depends on how decisively we act today.

Affected young date palms PC: Authors

IE Editors add: Understanding the spread of red palm weevil (RPW) across India, both via infested planting materials and through adult beetle dispersal, is essential for developing region-specific management strategies in anticipation of future outbreaks across the Indian subcontinent.

For more details contact:
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IE Blog No. 289
All IE blogs are available on website
https://insectenvironment.com

Insect Environment Blog

A Native Entomopathogenic Fungus Breakthrough: *Aspergillus* sp. R55 for Managing Rugose Spiraling Whitefly in Oil Palm

8 March 2026

A.R.N.S. Subbanna and K. Suresh

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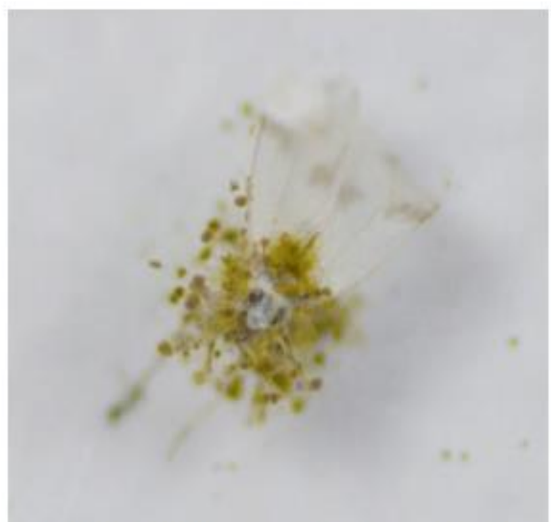
The globalization of agricultural trade has accelerated the spread of invasive pests, often overwhelming native ecosystems and causing steep economic losses for growers. One such pest, the Rugose Spiraling Whitefly (RSW), *Aleurodicus rugioperculatus*, has emerged as a major threat to India's plantation crops particularly oil palm, coconut, guava, and custard apple since its first detection in 2016. Its rapid proliferation, high reproductive capacity, and heavy honeydew excretion have created an urgent need for safe and sustainable control strategies.

Fungal mycosis on nymphs PC: Authors

Because RSW is an introduced (invasive) pest, the use of broad-spectrum chemical pesticides can disrupt native natural enemies and worsen outbreaks. Recognizing this challenge, scientists from the ICAR–Indian Institute of Oil Palm Research (IIOPR), Pedavegi explored native entomopathogenic fungus as eco-friendly solutions within the oil palm ecosystem. This exploration led to the discovery of a powerful indigenous biocontrol agent: *Aspergillus* sp. R55.



Fungal mycosis on adults PC: Authors

Laboratory assays revealed that R55 exhibits high pathogenicity against both adult and nymphal stages of RSW, with LT_{50} values of 1.5 and 2.04 days at 10^6 conidia/ml demonstrating rapid action. In two-year multilocational field trials across Andhra Pradesh (2023–2024), the fungus consistently reduced pest populations by 61.98% at 10 days and 94.44% at 15 days after application. Under humid conditions, R55 was capable of triggering natural epizootics, spreading infection within whitefly populations.

The isolate shows excellent mass-production potential (10^9 conidia/ml) and thermal tolerance, with over 50% conidial survival at 50°C for one hour an important trait for field stability in tropical climates. Ecotoxicological assessments confirmed R55's safety to mammals, birds, bees, earthworms, fish, and algae, supporting its suitability for large-scale integrated pest management.

This technology along with a two-step mass multiplication of the fungus (also developed at ICAR-IIOPR, Pedavegi) has now been commercialized to two firms in Andhra Pradesh, enabling production and distribution for wider use. Currently, R55 is deployed across ~2,900 acres in Eluru, West Godavari, East Godavari, Visakhapatnam, Srikakulam, and Vizianagaram, delivering significant reductions in RSW damage and providing growers with a sustainable, ecosystem-friendly alternative to chemical insecticides.

IE editors add: We appreciate the excellent work done by the authors in developing a high-temperature-resilient entomopathogenic fungus. It will be interesting to examine whether this strain performs equally well on other palm systems, offering broader biocontrol potential. This is a promising direction for future exploration.

IE Blog No. 295

All IE blogs are available on website https://insectenvironment.com

Field level epizootics caused by *Aspergillus* sp. R55 on adults of RSW PC: Authors

IE Extension

With Pomegranate farmers at Ittasandra, **Hoskote taluk** of Bangalore Rural district, Karnataka, India

Dr. M A Rashmi invited as a Special Guest for the Republic Day Celebrations 2026 at Kartavya Path, New Delhi by DBT–BIRAC

Team with Dr. Jitender Kumar, MD, BIRAC at New Delhi on 26th January 2026

With Dr. Arshi Mehboob, DD, BIRAC at New Delhi on 26th January 2026

With the students and staff of Biotechnology Department, UAS, GKVK, Bengaluru

Dr. Abraham Verghese delivering a special lecture on Biomanufacturing and commercialization for sustainable pest management in horticulture at Biotechnology Department, UAS, GKVK, Bengaluru on 19th February 2026

Dr. Abraham Verghese graced the Graduation Day at St. Thomas School, Bidadi as the Chief Guest on 6 March 2026

Dr. M. A. Rashmi, Phytosanitary Service Provider (PSSP) at Rashvee International Phytosanitary Research & Services (R-IPRS), supporting exporters with export-grade rose shipments during the Valentine season

Dr. Abraham Verghese delivering invited lecture at International Conference
Advances in Digital Solutions and Green Technologies in Crop Pest Management

With participants of International Conference at the Indian Statistical Institute (ISI) in Kolkata
from February 25 to 27, 2026

Dr. Abraham Verghese being felicitated by Dr. S. Jha at the Indian Statistical Institute (ISI) in Kolkata

Dr. Aabraham Verghese with Dr. M.R. Khan and Dr. C. Chatopdayaya at the Indian Statistical Institute (ISI) in Kolkata

With Dr. D Mandal in ISI campus, Kolkota

Dr. Abraham Verghese and V Susan with Dr. Ashok Kumar Patra, Vice Chancellor, Bidhan Chandra Krishi Viswavidyalaya (BCKV) in Mohanpur, Kalyani, West Bengal,

Dr. S. Deepak, Associate Editor, Insect Environment, interacting with government primary school children at Iraksandra, Gubbi Taluk, Tumkur, highlighting the importance of environmental conservation

Team participated in Farmer Producer Organizations (FPO's) & Startups Meet at UAS Dharwad, organized by KRISHIK Agri Business Incubator, Dharwad.

Rashvee Team with Mrs. Deepa Vastrad and Mrs. Manjula Laxmeshwar, KRISHIK Agri Business Incubator, Dharwad

With organizers at the *National Conference on Floriculture and Landscaping @2047 Viksit Bharat*, scheduled from 23–25 March 2026 at the College of Agriculture, KSNUAHS, Shivamogga

Dr. Abraham Verghese delivering the Keynote Address on ‘Emerging Precision Plant Protection Strategies in Floriculture for Enhanced Compliance Across Export and Domestic Markets,’ and being felicitated by the organizers at the National Conference on Floriculture and Landscaping @ 2047 Viksit Bharat, held from 23–25 March 2026 at the College of Agriculture, KSNUAHS, Shivamogga