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Special Issue: A Research Review on the Cotton Jassid *Amrasca biguttula*

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The Global Menace of the Indian Cotton Jassid: A Call for Sustainable Defense

*In an era of interconnected global agriculture, the Indian cotton jassid, *Amrasca biguttula* (Ishida, 1913), has earned its status as a true “global citizen”—a diminutive insect with an outsized capacity for devastation. Once confined to its Indo-Malayan origins, this polyphagous pest has now infiltrated cotton-growing regions worldwide, from the dry fields of West Africa to the Caribbean islands and the southeastern United States. For over a century, it has plagued Asia and Oceania, spanning from Iran to Japan and Micronesia, where it feeds on a broad array of economically vital crops like cotton, okra, eggplant, cowpea, potato, maize, sorghum, sunflower, pigeon pea, and beans. Its sap-sucking behavior induces “hopperburn,” a toxic condition that curls, yellows, and necroses leaves, stunting plants and slashing yields by up to 67%—or even causing complete crop failure if seedlings are targeted early. In West Africa alone, where the pest exploded in 2022, annual economic losses average US\$122 million, as calculated from seed cotton purchase prices, underscoring its threat to livelihoods in developing economies (WTO, 2024).*

*The jassid’s history is a tale of insect escapades and unintended consequences. Archaeological evidence reveals that farmers in the Indian subcontinent cultivated resilient diploid cotton species—*Gossypium arboreum* and *G. herbaceum* (Desi cottons)—for 3,000–4,000 years, coexisting with pests like the jassid without catastrophic losses. Yet, in 1790, the British East India Company introduced long-staple tetraploid species (*G. hirsutum* and *G. barbadense*) to fuel Britain’s textile mills, as Desi fibers were incompatible with new spinning machines. Acclimatization efforts failed for decades, with trials in Lyallpur (now Pakistan), Surat, Coimbatore, Lucknow, and Indore yielding limited success. A breakthrough came in 1912 when D. Milne developed varieties 3F and 4F; the glabrous 3F succumbed to jassids, but the hairy 4F survived. By 1933, Sardar Bahadur Labh Singh’s selection from 4F—LSS—became a Punjab landmark, maturing in 270 days and spreading rapidly. Subsequent varieties like 320F, J34 (190 days), and J205 (synchronous maturity) were hairy and tolerant, displacing Desi cottons in parts. Post-1947 independence, Indian breeders mandated jassid tolerance in *G. hirsutum* varieties, as susceptible lines required chemical protection to survive, highlighting the pest’s role in shaping modern cotton breeding.*

*Today, this legacy warns against complacency. New jassid invasions—in Iraq (2017), West Africa (2021–2022), Puerto Rico (2023), and the U.S. (101 counties by October 4, 2025)—demand proactive strategies, lest history repeat. Farmers often turn to insecticides as a first resort, but this “pesticide treadmill” fosters resistance to pyrethroids, organophosphates, and neonicotinoids (up to 1198-fold for thiamethoxam and 218-fold to imidacloprid), leading to resurgence and ecological collapse. Early-season applications, when jassids target seedlings, are particularly detrimental, killing beneficial predators and parasitoids like *Chrysoperla carnea*, lady beetles, and *Anagrus* spp., derailing season-long biological control and necessitating repeated sprays. India’s 1980s–1990s cotton crises, where overuse caused bollworm resurgence and farmer distress, stand as cautionary tales.*

*Sustainable management must prioritize ecofriendly methods. Start with jassid-resistant, hairy varieties like KC 2, NDLH 1938, or RAH 100, which deter oviposition and feeding through mechanisms such as trichomes, dense gossypol glands, biochemical and anatomical features, laying a firm IPM foundation by avoiding early disruptions. Seed treatment with neonicotinoids provides initial tolerance without broad-spectrum harm, while early sowing misaligns crop growth with peak jassid activity. Avoid excessive nitrogen to prevent succulent, attractive plants, and ensure potassium sufficiency to bolster resistance against hopper-burn. Weed removal and field sanitation eliminate overwintering sites, while crop rotation with non-hosts and avoiding intercropping with okra or eggplant break pest cycles. Initial reliance on botanical or microbial pesticides preserves natural enemies for season-long control. Research cited in this review shows that neem-based products could achieve 58–77.91% reduction, garlic extracts up to 54%, and seaweed tannins (*Sargassum wightii*, LC50 0.044%) disrupt enzymes without toxicity. Microbial agents like *Beauveria bassiana* (75.7% mortality, LT50 6.01 days) and *Purpureocillium lilacinum* (72.87% field reduction) target jassids selectively. If chemicals are needed (ETL exceeded), select safe options like flonicamid, dinotefuran, flupyradifurone, afidopyropen, sulfoxaflor, fluxametamide or biorationals to minimize non-target effects without broad-spectrum toxicity to the ecosystems. Precision tools like satellite imagery for stress detection enable targeted interventions, reducing chemical use. In the U.S., avoiding highly hazardous pesticides (HHPs) like Bidrin (dicrotophos, WHO Class Ib, LD50 17 mg/kg) is imperative—its early-season broad-spectrum toxicity kills pollinators and predators, risking outbreaks and resistance without superior efficacy over safer alternatives. U.S. extension bulletins recommending Bidrin overlook these risks, potentially derailing IPM.*

*This special issue consolidates knowledge on *A. biguttula*’s biology, ecology, distribution, impacts, and management, identifying gaps in invasion dynamics and host interactions to guide sustainable strategies. Global collaboration is essential to combat this “global citizen” before it exacts further tolls.*

– Keshav Kranthi

The Indian Cotton Jassid, *Amrasca biguttula* (Ishida, 1913): A Review

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an MSc with gold medals (1987), and a BSc (1985) from PJTSAU, she progressed from Scientist (1991) to Principal Scientist (2008–2020) and Head of Division (2011–2020) at ICAR-Central Institute for Cotton Research, Nagpur. Her accolades include the 2008 Outstanding Woman Scientist Award and multiple fellowships. Kranthi has led transformative projects, such as AI-GPS soil health apps, VR IPM trainings across Africa, and RNAi pest management innovations, yielding 28% yield increases and 50–90% pesticide reductions. She has authored 60 papers, 6 book chapters, and registered two Bt cotton varieties, submitting 52 gene sequences to GenBank.

Introduction

The Indian cotton jassid, *Amrasca biguttula* (Ishida, 1913) (Hemiptera: Cicadellidae: Typhlocybae), also known as *A. devastans* or *A. biguttula biguttula*, is a highly polyphagous sap-sucking pest commonly referred to as the okra leafhopper, cotton leafhopper, two-spot cotton leafhopper, or green jassid (Cabrera-Asencio *et al.*, 2023; Kshirsagar *et al.*, 2012). This pest has recently gained attention due to its invasion into new territories, including Iran, Iraq, West Africa, Puerto Rico, and the United States, expanding beyond its native range in Asia and Oceania, from Iran to Japan and Micronesia (Al-Hamadany & Al-Karboli, 2017; Cabrera-Asencio *et al.*, 2023; Dumbardon-Martial & Pierre, 2025). These invasions are likely facilitated by global trade, climate change, and the pest's adaptability to diverse agroecosystems, posing emerging threats to agricultural productivity in subtropical and tropical regions worldwide (Cabrera-Asencio *et al.*, 2023).



Image source: ICAR-CICR

Figure-1 Adult *Amrasca biguttula*

Morphologically, *A. biguttula* adults are small (approximately 2 mm), pale green insects with translucent wings featuring two distinct black spots, one on each wing tip, which aid in distinguishing them from similar species, particularly in the adult stage compared to nymphs (Cabrera-Asencio *et al.*, 2023). Both adults and nymphs feed primarily on the undersides of leaves, injecting salivary toxins that induce “hopper-burn,” a phytotoxic condition characterized by yellowing, leaf curling, browning, and necrosis (Ahmad *et al.*, 1985). Infestation symptoms often first appear along field edges and may be mistaken for potassium deficiency, but severe cases result in leaf abscission, stunted plant growth, and significant yield reductions (Ahmad *et al.*, 1985; Bhat *et al.*, 1986). If disturbed, the insects exhibit rapid sideways movement or hopping behavior and remain active throughout the year, with heightened activity during hot summer periods (Khaing *et al.*, 2002). Eggs are laid in soft plant tissues, enabling continuous population buildup across seasons (Jaysimha *et al.*, 2012).

As a key pest in the Indian subcontinent since the early twentieth century, *A. biguttula* attacks a wide range of economically important crops and wild hosts, including cultivated and wild cotton (*Gossypium* spp.), eggplant (*Solanum melongena*), potato (*Solanum tuberosum*), okra (*Abelmoschus esculentus*), cowpea (*Vigna unguiculata*), maize (*Zea mays*),

sunflower (*Helianthus annuus*), beans, sorghum, pigeon pea (*Cajanus cajan*), China rose (*Hibiscus rosa-sinensis*), and various grasses such as durva (*Cynodon dactylon*) (Dung *et al.*, 2021; Kamble & Sathe, 2015; Murugesan & Kavitha, 2010).

A. biguttula has caused devastating losses to cotton (*Gossypium hirsutum*), ranging from 37–67% in yield, with complete crop failure reported in localized areas; specific estimates include 100–114 kg of fiber per hectare.

This review synthesizes published literature up to the end of September 2025 to consolidate current knowledge on *A. biguttula*, highlighting research findings on its biology, ecology, distribution, impacts and management. By identifying gaps in understanding, it aims to guide future investigations into understudied aspects of the pest's life cycle, invasion dynamics, and host interactions, while informing the development of sustainable management strategies to mitigate its global agricultural threats.

Symptoms of Damage

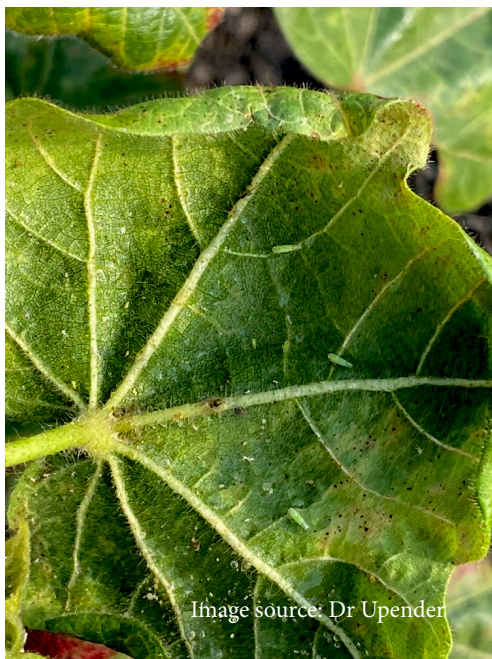
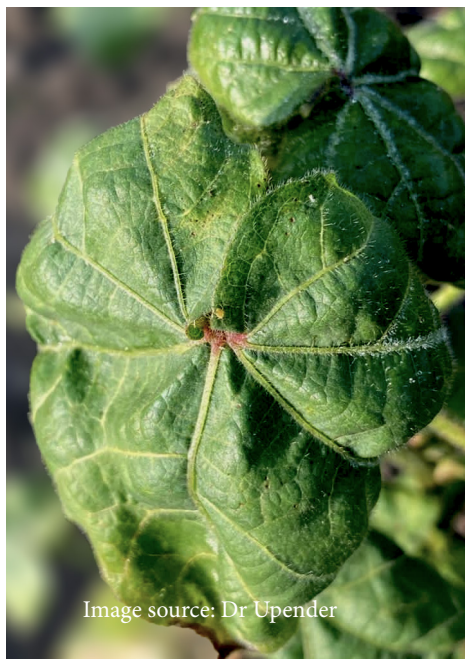


Figure-2. Leaf damage inflicted by *Amrasca biguttula* results from its sap-sucking behavior, primarily targeting the undersurface of leaves, which initiates a characteristic downward curling along the edges. As the infestation advances, the affected areas transition from yellowish hues to reddish tones, eventually encompassing the entire leaf in a mottled, red appearance before it withers and drops.

Symptoms of Damage

A. biguttula, causes extensive damage to host plants through its sap-sucking feeding behavior, primarily targeting the undersides of leaves (Dung *et al.*, 2021; Schreiner, 2000). Both nymphs and adults employ piercing-sucking mouthparts to extract sap from mesophyll cells and, in some instances, phloem tissues, injecting toxic saliva that induces a condition known as “hopper-burn” (Kamble & Sathe, 2015; Rajendran *et al.*, 2018; Reddy *et al.*, 2020). This toxemia results in initial

symptoms such as pale green to yellow blotches, often with a characteristic yellow “halo” along leaf edges, which can be mistaken for nutrient deficiencies like potassium deficiency (AVRDC, 2003; Belachew *et al.*, 2024; Schreiner, 2000). As feeding progresses, leaves exhibit downward curling, crinkling, marginal rolling, and chlorosis, followed by reddening, bronzing, and eventual browning or brick-red discoloration (Butani & Jotwani, 1983; Meena *et al.*, 2010; Tayyab *et al.*, 2024). These symptoms typically begin at field edges and spread inward, complicating early detection (Schreiner, 1990).

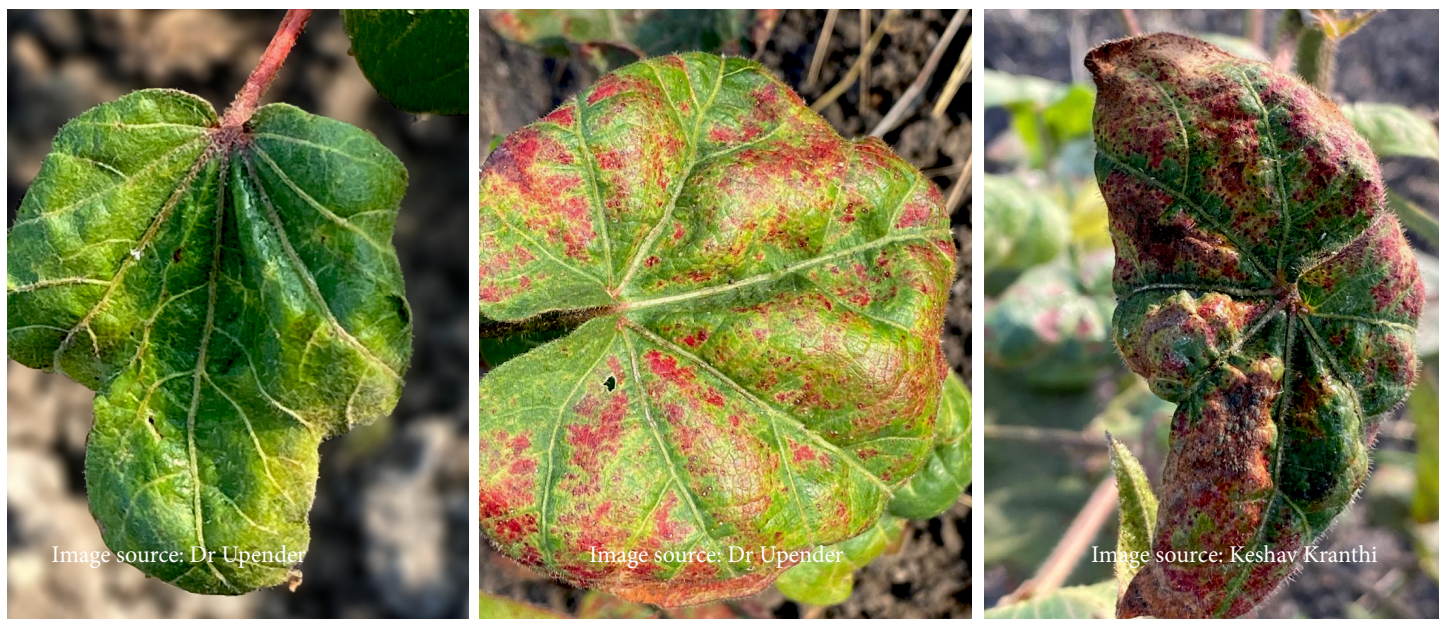


Figure-2a. Damage symptoms in cotton leaves.

Under severe infestations, the damage intensifies, leading to necrosis of leaf tips and margins, brittleness, and defoliation, with plants experiencing stunted growth -especially if jassids infest the crop in early stages, reduced boll production, and premature shedding of buds, flowers, and fruiting bodies, thereby significantly impacting crop yields (Dung *et al.*, 2021; Ghosh & Karmakar, 2022; Madar *et al.*, 2010; Narayanan & Singh, 1994; Rehman, 1940). In cotton (*Gossypium hirsutum*), heavy jassid populations can cause leaves to dry out and die, particularly in the lower canopy, with severe cases resulting in plant death (Madar *et al.*, 2010; Tayyab *et al.*, 2024). Additionally, the secretion of honeydew during feeding promotes the growth of sooty mold, a fungal layer that further impairs photosynthesis and reduces plant vitality (Kamble & Sathe, 2015; Rajendran *et al.*, 2018). The similarity of hopper-burn symptoms to those caused by spider mite infestations or vascular plant pathogens or potassium deficiency, underscores the need for accurate scouting to identify *A. biguttula* as the causative agent, ensuring appropriate management interventions (Schreiner, 2000; Reddy *et al.*, 2020). The cumulative effects of these symptoms highlight the destructive potential of *A. biguttula* across crops such as cotton, eggplant, okra, and sunflower, necessitating vigilant monitoring and control strategies (Belachew *et al.*, 2024; Dung *et al.*, 2021).

Yield Losses

A. biguttula causes significant economic losses across multiple crops, particularly cotton and okra in South Asia and Africa. While yield losses in the pest’s native range (Iran to Japan and Micronesia) have not been precisely quantified, estimates suggest feeding damage can cause >60% reductions in cotton yields (Ahmed, 1982; Ahmad *et al.*, 1985). In India, yield losses in cotton were 44% due to bollworms, 12% from sucking pests, and 52% from both combined (Dhawan *et al.*, 1988). Jassids caused yield reductions exceeding 100–114 kg lint/ha in cotton during peak summer infestations (Sukhiya *et al.*, 1987; Dhawan *et al.*, 1988). On transgenic cotton, yield loss estimates range between 13.13% (tolerant hybrids such as Tulasi-9NBt) and 19.99% (susceptible hybrids such as RCH-2NBt) (Ramalakshmi *et al.*, 2019). Earlier, Ahmad *et al.* (1986) reported 20–40% yield loss due to stunting and plant mortality in early cotton. Losses attributed to sucking pest complexes in cotton have been reported at 5–45% (Ahuja *et al.*, 2009).

In other crops, impacts are equally severe. Yield losses are particularly high in okra, with reports of ~50% reduction (Devi *et al.*, 2018), while eggplant may suffer up to 37% yield losses (Ahmed, 1982). Sunflower and roselle have also been

Table-1. Estimates of yield loss caused by *A. biguttula*

Host Crop	Location	Yield Loss Estimate	Notes / Observations	Reference(s)
Cotton	India	12% (sucking pests), 44% (bollworms), 52% (combined)	North India	Dhawan <i>et al.</i> , 1988
Cotton	India	20–40%	Stunting and plant mortality	Ahmad <i>et al.</i> , 1986
Cotton	India	>100–114 kg lint/ha	About 20% yield loss	Sukhija <i>et al.</i> , 1987; Dhawan <i>et al.</i> , 1988
Cotton	India	13.13% (Tulasi-9NBt), 19.99% (RCH-2NBt)	RCH-2 more susceptible; Tulasi-9 more tolerant	Ramalakshmi <i>et al.</i> , 2019
Cotton	India	5–45%	Range of losses reported	Ahuja <i>et al.</i> , 2009
Cotton	Iran to Japan, Micronesia	Up to >60%	Feeding damage estimates	Ahmed, 1982; Ahmad <i>et al.</i> , 1985
Cotton	Senegal	38% reduction in exports	Significant financial impact,	PR-PICA, 2022; Syl-la, 2023
Cotton	Côte d'Ivoire	XOF 34 billion (USD 59 million) loss	Recent outbreak	Kouadio <i>et al.</i> , 2024
Cotton	Burkina Faso	XOF 65 billion loss (USD 105 million)	Recent outbreak	Kouadio <i>et al.</i> , 2024
Okra	India	~50%	Substantial crop loss	Devi <i>et al.</i> , 2018
Okra	South Asia	Severe reductions (not quantified)	Major cause of reduced production	Ghosh and Karmakar, 2021; Saeed <i>et al.</i> , 2015
Eggplant	India	37%+	Yield loss increases with pest density	Ahmed, 1982
Eggplant	South Asia	Significant market losses (not quantified)	Reduced production and trade	Ghosh and Karmakar, 2021; Saeed <i>et al.</i> , 2015
Sunflower	India	Not quantified (severe)	Yield losses observed	Madar and Katti, 2011; Manivannan <i>et al.</i> , 2021
Roselle	India	Not quantified (severe)	Crop damage not-ed	Manivannan <i>et al.</i> , 2021
Grapes	India & Phillippines	Lesser extent (not quantified)	Economic impacts lower than cotton/okra	Lit & Bernardo, 1990; Schreiner, 1990; Ghosh & Karmakar, 2021
Multiple	Global	>50% under favorable pest conditions	Cotton, okra, sun-flower most impacted	Cabrera-Asencio <i>et al.</i> , 2023

noted as vulnerable hosts, with substantial but less quantified yield reductions (Madar and Katti, 2011; Manivannan *et al.*, 2021). Grapes and other horticultural crops can be affected to a lesser extent (Lit and Bernardo, 1990; Schreiner, 1990; Ghosh and Karmakar, 2021). Yield losses exceeding 50% in certain crops under favorable pest conditions have also been documented (Cabrera-Asencio *et al.*, 2023).

In Africa, jassid outbreaks are emerging as a major threat to cotton and other crops. Kouadio *et al.* (2024) estimated economic losses of XOF 34 billion (USD 59 million) in Côte d'Ivoire and XOF 65 billion (USD 105 million) in Burkina Faso during recent infestations. According to a WTO report (2024), the cotton sector in West Africa has suffered significant setbacks due to *Amrasca biguttula* infestations, with devastating effects on production and yields. In Senegal, output and yields plummeted by 48% and 40%, respectively, compared to projections at the onset of the 2022-23 season, while Togo experienced a notable 10% drop in production.

Côte d'Ivoire faced particularly severe damage, with crop losses reaching 41% and yield reductions hitting 50%, driven by intense pest pressure. In Mali, Benin, and Burkina Faso—collectively contributing about half of the region's cotton output—farmers and officials have reported an unprecedented surge in jassid activity, amplifying the crisis. Across the six affected West African nations, crop losses for the 2022-23 season ranged from 8% to 50%, with Côte d'Ivoire losing an estimated 236,186 tonnes of seed cotton, a figure derived from its three-year average yield prior to the outbreak, translating to a staggering economic toll of 73,217,660,000 CFA francs based on purchase prices. Extrapolating these losses across all eight PR-PICA countries suggests a regional impact nearing 215 billion CFA francs, underscoring the profound economic strain this pest imposes on cotton farming. Market-level disruptions have been noted, including a 38% reduction in cotton exports from Senegal, with governments forced to implement subsidies and loan forgiveness programs to offset farmer losses (PR-PICA, 2022; Syl-la, 2023).

Overall, *A. biguttula* has emerged as one of the most noxious pests of cotton and okra, with yield losses ranging from 12% to >60% depending on crop, genotype, and agro-ecological conditions (Navasero, 2015). In addition to direct yield reductions, trade implications may arise if infestations establish in new regions such as the United States, where cotton, grapes, and eggplant are economically important

Disease Vector Potential

A. biguttula, has been implicated as a potential vector of plant pathogens, particularly phytoplasmas and viruses, but its role remains unconfirmed (Biswas *et al.*, 2018; Nielson, 1968; Reddy *et al.*, 2020; Sharif *et al.*, 2019). Historical reports from India suggested that

A. biguttula (previously identified as *Empoasca devastans*) might transmit phytoplasmas, initially mistaken for viruses, in crops such as cotton and other hosts (Hill, 1943; Nielson, 1975; Thomas & Krishnaswami, 1939). However, these early claims lack substantiation due to limited experimental evidence and outdated taxonomic classifications.

More recent studies have associated *A. biguttula* with phytoplasma species, including those linked to chickpea chlorotic dwarf virus and other phytoplasma-related diseases in crops like sunn hemp (*Crotalaria juncea*) and vegetables (Biswas *et al.*, 2018; Reddy *et al.*, 2020; Sharif *et al.*, 2019). Despite these associations, no conclusive evidence confirms *A. biguttula* as a competent vector for these pathogens. The damage caused by *A. biguttula*, such as hopper-burn, often mimics symptoms of vascular plant pathogen infections, including chlorosis, leaf curling, and necrosis, which may contribute to misattributions of disease transmission (Reddy *et al.*, 2020).

The absence of definitive vector studies highlights a critical knowledge gap, necessitating further research to clarify the pest's role in pathogen transmission and its broader implications for crop health.

History of *Amrasca biguttula* as a Pest

The Indian cotton jassid, *Amrasca biguttula*, historically referred to as *Empoasca devastans* or *Chlorita flavescens*, has been recognized as a significant agricultural pest since the early twentieth century, particularly in regions with favorable climatic conditions for its proliferation.

A. biguttula has been recognized as a significant pest of cotton, okra, brinjal, and numerous other crops across Asia and Southern Africa for over a century, with its impact documented since the early 20th century (e.g., Ishida, 1913a; Maxwell-Lefroy, 1906a). This polyphagous leafhopper, known for causing hopper-burn through sap-sucking, has adapted to diverse agroecosystems, thriving in warm, humid conditions typical of these regions. Its long history of infestation, historically spanning mainly from India and Pakistan to parts of

East Africa, and its recent invasion into the west, reflects its resilience and ability to exploit a wide range of hosts, posing a persistent challenge to agricultural productivity.

Early reports from South Africa during the 1922–23 cotton season documented widespread damage by jassids, with infestations noted across the Transvaal, Zululand, and Swaziland, attributed to abnormal weather conditions characterized by excessive rainfall and cloudy weather (Worrall, 1923). These conditions facilitated rapid population growth, leading to significant yield losses across the Union of South Africa, with symptoms including yellowing, reddening, and drying of leaves, though not necessarily leaf shedding (Worrall, 1923). By 1925, continued heavy rainfall in the Eastern Transvaal lowveld exacerbated jassid damage, reducing cotton yields by at least 25% in most areas, with some regions experiencing even greater losses (Worrall, 1925). These early accounts highlighted the pest's sensitivity to environmental factors, such as high humidity and prolonged wet periods, which promoted its spread and intensified its impact on cotton crops (Worrall, 1923, 1925).

In the Indian subcontinent, *Amrasca biguttula* has posed a persistent threat to American cotton varieties (*Gossypium hirsutum*) cultivation for over a century, challenging farmers who have grown the diploid species *Gossypium arboreum* and *Gossypium herbaceum* (Desi cottons) for 3,000–4,000 years, as evidenced by archaeological records. In 1790, the British East India Company introduced the long-staple tetraploid species *Gossypium hirsutum* and *Gossypium barbadense* to replace these short-staple varieties, aiming to supply the Manchester and Lancashire textile industries with cotton suited for new spinning machines. Initial efforts to acclimatize these species across India—through trials in Lyallpur, Shahpur (now Pakistan), Surat, Coimbatore, Lucknow, and Indore—met with limited success for decades. Breakthrough came in 1912 when D. Milne, using seeds provided in 1853, developed the varieties 3F and 4F in Punjab; while the glabrous 3F succumbed to jassid attacks, the hairy 4F showed resilience, though its adaptation was modest. In 1933, Sardar Bahadur Labh Singh's selection from 4F, named LSS, marked a turning point in Punjab's cotton history, spreading rapidly despite its 270-day maturity. This led to the development of 320F (a faster-maturing LSS derivative), followed by J34 (190 days) and J205 (synchronous maturity), both hairy and jassid-tolerant, gradually displacing Desi cottons.

Special research focusing on jassids was initiated in 1937 at Lyallpur (now Faisalabad, Pakistan) to address the threat to the adaptability of the species *G. hirsutum* (Afzal & Ghani, 1953). Studies revealed that jassids infestations typically begin six weeks after sowing in late June or early July, peaking in August and declining by November, with high atmospheric humidity identified as the primary factor driving population increases (Afzal & Ghani, 1953). The pest's preference for oviposition and feeding on a wide range of host plants, including okra (*Abelmoschus esculentus*), eggplant (*Solanum melongena*), potato (*Solanum tuberosum*), sunflower (*Heli-*

anthus annuus), and various *Hibiscus* species, highlights its polyphagous nature (Afzal & Ghani, 1953). Severe infestations in 1944 reduced yields of susceptible cotton varieties to nearly a quarter of those in 1942, when jassid populations were minimal, demonstrating the pest's devastating impact on boll production (Afzal & Ghani, 1953).

Notably, early research identified leaf hairiness as a key factor in varietal resistance, with hairy cotton varieties showing reduced oviposition success, a finding that laid the groundwork for breeding resistant cultivars (Afzal & Ghani, 1953). These historical observations established *A. biguttula* as a persistent and economically significant pest, with its impact amplified by environmental conditions and host availability, setting the stage for subsequent research into its biology and management.

Post-1947 independence, Indian breeders prioritized developing *G. hirsutum* varieties with inherent jassid tolerance, as susceptible types required chemical seed treatments and struggled to establish without early-season protection, underscoring the pest's enduring influence on cotton breeding.

Taxonomy

The Indian cotton jassid, *Amrasca biguttula* (Ishida, 1913) (Hemiptera: Cicadellidae), is a member of the tribe Empoascini within the subfamily Typhlocybinae, the second largest tribe of microleafhoppers comprising approximately 1,372 species worldwide (Xu *et al.*, 2017).

The taxonomy of *A. biguttula* is complex due to its historical classification under multiple genera and species names, reflecting challenges in morphological identification and taxonomic revisions.

Originally described as *Chlorita biguttula* by Ishida (1913), the species has been synonymized with several names, including *Empoasca biguttula* (Shiraki, 1913), *Empoasca devastans* (Distant, 1918), *Chlorita bimaculata* (Matsumura, 1916), *Empoasca bipunctata* (Schumacher, 1915), *Empoasca nigropunctata* (Merino, 1936), *Empoasca quadrinotatissima* (Dlabola, 1957), *Empoasca uniguttata* (Jacobi, 1941), and *Zygina biguttula punctata* (Melichar, 1914), among others (Dworakowska, 1970; Sohi, 1983; Xu *et al.*, 2017).

The subspecies *Amrasca biguttula biguttula* and *Amrasca biguttula punctata* have also been recognized, with the latter considered a subjective synonym of *Chlorita biguttula* (Dworakowska, 1976; Xu *et al.*, 2017). The genus *Sundapteryx* was briefly used for this species (e.g., *Sundapteryx biguttula*), but *Amrasca* is now the accepted genus following taxonomic revisions (Dworakowska & Viraktamath, 1975; Xu *et al.*, 2017).

Accurate identification of the *A. biguttula*, is critical due to its morphological similarity to other leafhoppers within the tribe Empoascini, such as *Empoasca* species (e.g., *E. fabae*, *E. vitis*, *E. kraemeri*), which are small, pale green insects

with yellowish-green, translucent wings, and approximately 2 mm in length, but lack the characteristic diagnostic black spots one near the apex of the brachial cell of each forewing and a pair of smaller spots pre-apically on the crown of the head, that distinguish *A. biguttula* adults (Cabrera-Asencio *et al.*, 2023; Esquivel *et al.*, 2025; Schreiner, 2000; Reddy *et al.*, 2020; Sagarbarria *et al.*, 2020). These markings, though variably developed and occasionally faded (especially head spots in older adults), allow differentiation from most native North American and Neotropical Empoascini, such as *Empoasca fabae* (potato leafhopper), *E. vitis*, and *E. kraemeri*, which lack these specific spots (Cabrera-Asencio *et al.*, 2023; Sagarbarria *et al.*, 2020).

Other Typhlocybinae, such as *Alconeura* spp., *Dikrella* spp., and *Eratoneura* spp., may have wing spots but are not primarily green, and genera like *Kyboasca* (Holarctic) or *Tripunctiasca* (Mexican) either lack head spots or have spots in different wing cells (e.g., second apical cell in *Alconeura*) (Cabrera-Asencio *et al.*, 2023; Dmitriev *et al.*, 2022). The expanding global distribution further complicates accurate identification to differentiate it from its related species (Cabrera-Asencio *et al.*, 2023; Xu *et al.*, 2017).

Nymphs are nearly indistinguishable from those of *Empoasca* species, necessitating examination of male genitalia or molecular characterization for precise identification (Reddy *et al.*, 2020; Sagarbarria *et al.*, 2020).

Nymphs -wingless and pale green, are highly agile, exhibiting rapid sideways movement when disturbed, but lack distinctive markings, making them nearly indistinguishable from nymphs of other leafhoppers like *Empoasca* spp. (Jayasimha *et al.*, 2012; Schreiner, 2000).

Later-instar nymphs may develop small black spots on the pro-, meso-, and meta-notum, providing limited diagnostic value (Cabrera-Asencio *et al.*, 2023). Both adults and nymphs feed on the undersides of leaves, stems, and petioles, producing honeydew that compromises plant health (AVRDC, 2003; Rajendran *et al.*, 2018).

Tentative field identification can be based on host damage (e.g., hopper-burn) and external markings, but definitive identification requires morphological examination of male genitalia, characterized by a pair of lateral apodemes extending into abdominal segment VI and tergum VIII with arched internal ridges (Esquivel *et al.*, 2025; Xu *et al.*, 2017).

Molecular barcode data, such as COI gene sequencing, can supplement identification when males are unavailable, though male voucher specimens are preferred for establishing new records (Kranthi *et al.*, 2018; Sagarbarria *et al.*, 2020).

The lack of species-specific lures or traps complicates monitoring, as visual surveys and yellow or green sticky traps capture non-target species, increasing the risk of misidentification with similar leafhoppers such as *Typhlocyba pomeria* or *Alconeura* spp. *Alebroides nigroscutulatus* and *Seriana equa-*

ta, which may be mistaken for *A. biguttula* without detailed morphological or genetic analysis (Sagarbarria *et al.*, 2020).

Historical taxonomic confusion, with *A. biguttula* often par-allely referred as *Amrasca devastans* or other synonyms in economic entomology literature, and cases of misidentifica-tions, such as *Empoasca devastans* (Maxwell-Lefroy, 1906), a nomen nudum due to the absence of a formal description, and misspellings like *Empoasca biguttala* or *Chlorita bigut-tula*, underscores the need for rigorous taxonomic studies and identification protocols to clarify synonymies and sta-bilize nomenclature (Dworakowska, 1970; Li *et al.*, 2005; Xu *et al.*, 2017).

The confusion with taxonomic identification highlights the need and importance of Type specimens for *A. biguttula* and its synonyms, including lectotypes deposited at institu-tions like the Entomological Institute, Hokkaido University, and holotypes at the National Museum of the Philippines,

provide critical reference points for taxonomic validation (Dworakowska, 1970; Xu *et al.*, 2017).

Recent advances in molecular techniques and standardized morphological protocols, including clearing male abdomens in 10% potassium hydroxide for genitalia examination, have improved diagnostic accuracy, particularly for confirming invasive populations in regions like Puerto Rico and the United States (Cabrera-Asencio *et al.*, 2023; Esquivel *et al.*, 2025).

These methods are essential for distinguishing *A. biguttula* from closely related species and supporting effective pest surveillance and management.

Further, the ongoing molecular phylogenetic studies, lever-aging techniques like COI gene sequencing, are essential to resolve remaining taxonomic ambiguities and confirm the species' identity across its expanding global range (Kranthi *et al.*, 2018; Sagarbarria *et al.*, 2020).

Table-2. Synonyms and homonyms of *Amrasca biguttula* (Ishida, 1913)

Synonym	Author and Year	Citation/Reference
<i>Amrasca (Amrasca) biguttula</i>	Ishida, 1913	Imran Khatri <i>et al.</i> , 2013
<i>Amrasca (Sundapteryx) biguttula</i>	Ishida, 1913	Xu <i>et al.</i> , 2017
<i>Amrasca biguttula</i>	Ishida, 1913	Dworakowska, 1977
<i>Amrasca biguttula biguttula</i>	Ishida, 1913	Dworakowska & Viraktamath, 1975
<i>Amrasca biguttula punctata</i>	Melichar, 1914	Dworakowska, 1976
<i>Amrasca devastans</i>	Distant, 1918	Ahmed & Samad, 1972
<i>Chlorita biguttula</i>	Ishida, 1913	Ishida, 1913
<i>Chlorita biguttula biguttula</i>	Ishida, 1913	Not explicitly cited; inferred from subspecies
<i>Chlorita biguttula</i>	Shiraki, 1913	Shiraki, 1913
<i>Chlorita bimaculata</i>	Matsumura, 1916	Matsumura, 1916
<i>Chlorita quadrinotatissima</i>	Dlabola, 1957	Dlabola, 1958
<i>Empoasca biguttula</i>	Ishida, 1913	Anonymous, 1936
<i>Empoasca biguttula</i>	Shiraki, 1913	Shiraki, 1913 (as per CABI)
<i>Empoasca bipunctata</i>	Schumacher, 1915	Schumacher, 1915
<i>Empoasca devastans</i>	Distant, 1918	Distant, 1918
<i>Empoasca devastans</i>	Maxwell-Lefroy, 1906	Maxwell-Lefroy, 1906
<i>Empoasca nigropunctata</i>	Merino, 1936	Merino, 1936
<i>Empoasca quadrinotatissima</i>	Dlabola, 1957	Dlabola, 1957
<i>Empoasca schumacheri</i>	Metcalf, 1968	Metcalf, 1968
<i>Empoasca uniguttata</i>	Jacobi, 1941	Jacobi, 1938
<i>Sundapteryx biguttula</i>	Ishida, 1913	Not explicitly cited; inferred from combinations
<i>Sundapteryx biguttula biguttula</i>	Ishida, 1913	Dworakowska, 1970
<i>Sundapteryx biguttula punctata</i>	Melichar, 1914	Dworakowska, 1970
<i>Typhlocyba uniguttata</i>	Jacobi, 1941	Metcalf, 1968
<i>Zygina biguttula punctata</i>	Melichar, 1914	Melichar, 1914

Table-3. Diagnosis of *Amrasca biguttula*

Description	Diagnostic Notes	Citation
Adult Morphology		
Small (2 mm), pale green with yellowish-green, translucent wings. Two black spots near the apex of each forewing (brachial cell) and a pair of smaller black spots preapically on the crown of the head.	Spots variably developed; head spots may fade in older adults. Distinguishes from <i>Empoasca</i> spp. (e.g., <i>E. fabae</i> , <i>E. vitis</i> , <i>E. kraemer</i>), which lack these spots.	Cabrera-Asencio <i>et al.</i> , 2023; Esquivel <i>et al.</i> , 2025; Schreiner, 2000; Xu <i>et al.</i> , 2017
Nymph Morphology		
Wingless, pale green, highly agile with rapid side-ways movement when disturbed. Later instars may show small black spots on pro-, meso-, and meta-notum.	Nearly indistinguishable from <i>Empoasca</i> spp. nymphs; lacks distinctive markings except in later instars.	Cabrera-Asencio <i>et al.</i> , 2023; Jayasimha <i>et al.</i> , 2012; Schreiner, 2000
Behavior		
Adults and nymphs feed on undersides of leaves, stems, and petioles, producing honeydew. Exhibit crawling, jumping, or short flights when disturbed.	Feeding causes hopperburn; honeydew promotes sooty mold. Behavior similar to other leaf-hoppers but aids field identification when paired with host damage.	AVRDC, 2003; Ghosh & Karmakar, 2022; Rajendran <i>et al.</i> , 2018; Schreiner, 2000
Male Genitalia		
Lateral apodemes extending into abdominal segment VI; tergum VIII with a pair of arched internal ridges.	Definitive identification requires dissection and examination by a Cicadellidae specialist.	Esquivel <i>et al.</i> , 2025; Xu <i>et al.</i> , 2017
Molecular Identification		
COI gene sequencing (molecular barcode data) can be used when males are unavailable.	Preferred for confirming new records if male specimens are absent, but male vouchers are ideal.	Kranthi <i>et al.</i> , 2018; Sagarbarria <i>et al.</i> , 2020
Field Diagnostic Methods		
Visual surveys and yellow/green sticky traps; tentative identification based on host damage (hopper-burn) and external black spots.	Non-specific traps capture non-target species, requiring confirmation via genitalia or molecular methods.	Sagarbarria <i>et al.</i> , 2020
Morphological Processing		
Male abdomens cleared in 10% potassium hydroxide (KOH), rinsed, and immersed in glycerine for genitalia examination.	Enhances accuracy for confirming invasive populations (e.g., Puerto Rico, USA).	Cabrera-Asencio <i>et al.</i> , 2023; Esquivel <i>et al.</i> , 2025
Easily Mistaken Species		
<i>Empoasca</i> spp. (<i>E. fabae</i> , <i>E. vitis</i> , <i>E. kraemer</i>): small, green, lack black spots on head/forewings. <i>Alconeura</i> spp., <i>Dikrella</i> spp., <i>Eratoneura</i> spp.: non-green with spots in different wing cells. <i>Kyboasca</i> spp.: black spots on forewings but not on head. <i>Tripunctiasca</i> spp.: head spots but no preapical wing spots. <i>Typhlocyba pome</i> ria: lacks black spots.	Morphological or molecular analysis needed to differentiate; <i>A. biguttula</i> spots in brachial cell (vs. second apical cell in <i>Alconeura</i>).	Cabrera-Asencio <i>et al.</i> , 2023; Dmitriev <i>et al.</i> , 2022; Sagarbarria <i>et al.</i> , 2020
Taxonomic Considerations		
Often misidentified as <i>Amrasca devastans</i> or other synonyms in economic literature. Correct name clarified as <i>Amrasca (Sundapteryx) biguttula</i> .	Historical confusion with synonyms necessitates rigorous taxonomic verification.	Xu <i>et al.</i> , 2017

Distribution

Historically *Amrasca biguttula* has exhibited a predominantly Asian and African distribution, with recent invasions into the Caribbean and North America, reflecting its adaptability to tropical and subtropical agroecosystems. *Amrasca biguttula* (Ishida, 1913), native to the Indo-Malayan region, has an established range across Asia and Oceania, stretching from Iran in the west to Japan, Taiwan, and Micronesia in the east, encompassing countries such as Afghanistan, Bangladesh, China, India, Indonesia, Laos, Myanmar, Nepal, Pakistan, the Philippines, Thailand, Vietnam, Sri Lanka, and the Pacific island of Guam (Dmitriev *et al.*, 2022; Fletcher *et al.*, 2017; Xu *et al.*, 2017). As a major pest of cotton, okra, and other crops for over a century, its presence spans the Indian subcontinent, Southeast Asia, and East Asia, where it thrives in diverse agroecosystems, causing significant damage to a wide range of crops.

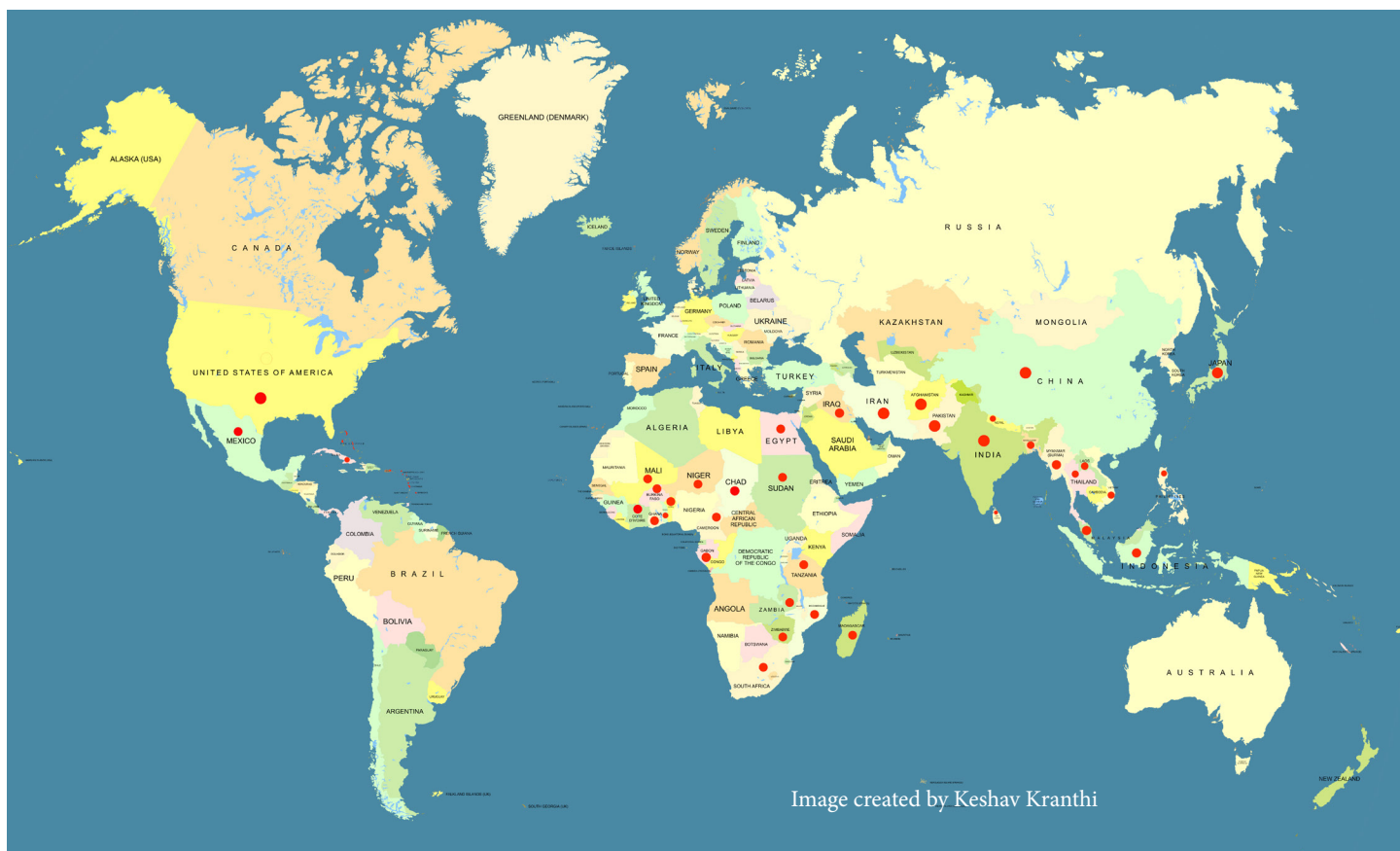


Figure-3. Global map of *Amrasca biguttula* distribution (red dots indicate the presence of *Amrasca biguttula* in the country)

Recent invasions have dramatically expanded *A. biguttula*'s global footprint, driven by international trade, climate suitability, and the absence of natural enemies in novel environments, aligning with the enemy release hypothesis (Middleton, 2008). The first recent indication of a westward movement appeared, a first record that emerged in Iraq in 2017 (Al-Hamdany & Al-Karboli, 2017). Though Taghizadeh *et al.* (2012) reported that *A. biguttula* was first detected in Iran in 2012, the pest may have been present in the region in earlier times (<https://plantwisepplusknowledgebank.org/doi/10.1079/pwkb.species.20857>). Though, *A. biguttula* was reported as a major pest of cotton from the South African region in 1922-23 (Worrall, 2023), and was known to be occur in Southern Africa (Madagascar, Mozambique, South Africa, Tanzania and Zambia), it was not reported from any part of West Africa for decades, but has established itself recently across West Africa and with emerging records in North Afri-

ca (Egypt). *Amrasca biguttula* invasion often displaces native leafhoppers leading to significant yield losses in cotton and vegetables (Jacques *et al.*, 2024; Kouadio *et al.*, 2024).

In West Africa *Amrasca biguttula* was first noted at low levels in Ghana as early as 1999 but surged in West Africa during 2022–2023, with confirmed outbreaks in Burkina Faso, Cameroon, Chad, Côte d'Ivoire, Benin, Ghana, Mali, Niger, Nigeria, Senegal, and Togo (Jacques *et al.*, 2024; Obeng-Ofori & Sackey, 2003; PR-PICA, 2022). By 2025, modeling projections indicate further range expansion across sub-Saharan Africa, potentially threatening cotton production in rainfed systems (Azrag *et al.*, 2025; Kouadio *et al.*, 2024). During the 2022-23 growing season, detailed morphological assessments across the six West African PR-PICA member nations—Benin, Burkina Faso, Côte d'Ivoire, Mali, Senegal, and Togo—revealed *Amrasca biguttula* as the prevailing jassid species, dominating over 90% of the population in these

regions (WTO, 2024). At that time, neither Cameroon nor Chad reported any impact from this outbreak. However, by the 2023-24 season, the pest's presence extended to all eight PR-PICA countries, highlighting its aggressive invasive tendencies and rapid spread.

In the Western Hemisphere, *A. biguttula* represents a novel invasive threat, with initial detection in southern Puerto Rico in April 2023, marking the first official record in the Americas (Cabrera-Asencio *et al.*, 2023). The entry into the Caribbean region includes Anguilla, Antigua and Barbuda, Bahamas, Barbados, Cuba, Mexico, Puerto Rico, Saint Kitts and Nevis, and U.S. Virgin Islands, and has since spread rapidly probably via trade and wind dispersal (Cabrera-Asencio *et al.*, 2023; Dmitriev *et al.*, 2024; EPPO, 2024; IPPC, 2024). In the United States, the pest was confirmed in Florida in November 2024, followed by infestations in multiple counties across Georgia, Alabama, and South Carolina by mid-2025, with detections in at least 13 Georgia counties since July 2024, posing risks to cotton, peanuts, soybeans, and ornamental nurseries (CAPS, 2025; Esquivel *et al.*, 2025; NC State Extension, 2025; Reisig & Collins, 2025). As of

September 2025, *A. biguttula* has not been confirmed in North Carolina, but its proximity and dispersal capabilities suggest imminent establishment in southeastern U.S. cotton belts (Reisig & Collins, 2025). Climate change models forecast heightened invasion risk in cotton-producing regions of South America, sub-Saharan Africa, and parts of the southern U.S. under future warming scenarios, emphasizing the need for enhanced surveillance and phytosanitary measures (Azrag *et al.*, 2025). The pest's adaptability to island ecosystems and lack of co-evolved predators in invaded ranges amplify its potential for unchecked proliferation, particularly on *Gossypium* hosts native to both hemispheres (Dmitriev *et al.*, 2022; Middleton, 2008). Although records suggest a single occurrence on Christmas Island (an Australian territory), the species is not considered established in mainland Australia, where the native *Amrasca terraereginae* predominates (Fletcher *et al.*, 2017). Other oceanic distributions include Guam, and French Polynesia, likely introduced via maritime trade (Fletcher *et al.*, 2017). As of September 30, 2025, no confirmed records exist for Rio de Janeiro (Brazil) or Botswana, Kenya, and Israel, highlighting gaps in surveillance that could facilitate further spread (Azrag *et al.*, 2025).

Host Plants

Amrasca biguttula, is a selectively polyphagous pest that exploits a diverse range of cultivated and wild host plants across multiple plant families, primarily Malvaceae, Fabaceae, Solanaceae, Asteraceae, and Poaceae (CABI, 2024; Saeed *et al.*, 2015). Its ability to feed on both agricultural crops and alternative hosts enables it to persist in agroecosystems year-round, acting as a reservoir for pest populations during periods when primary hosts like cotton (*Gossypium hirsutum*)

are unavailable, thereby facilitating migration and severe infestations (Clementine *et al.*, 2005; Huque, 1994).

The presence of alternative hosts can also influence pest dynamics by harboring natural enemies, though intensive insecticide use may disrupt these interactions, indirectly affecting pest populations on nearby crops (Edwards *et al.*, 1990; Naveed *et al.*, 2007).

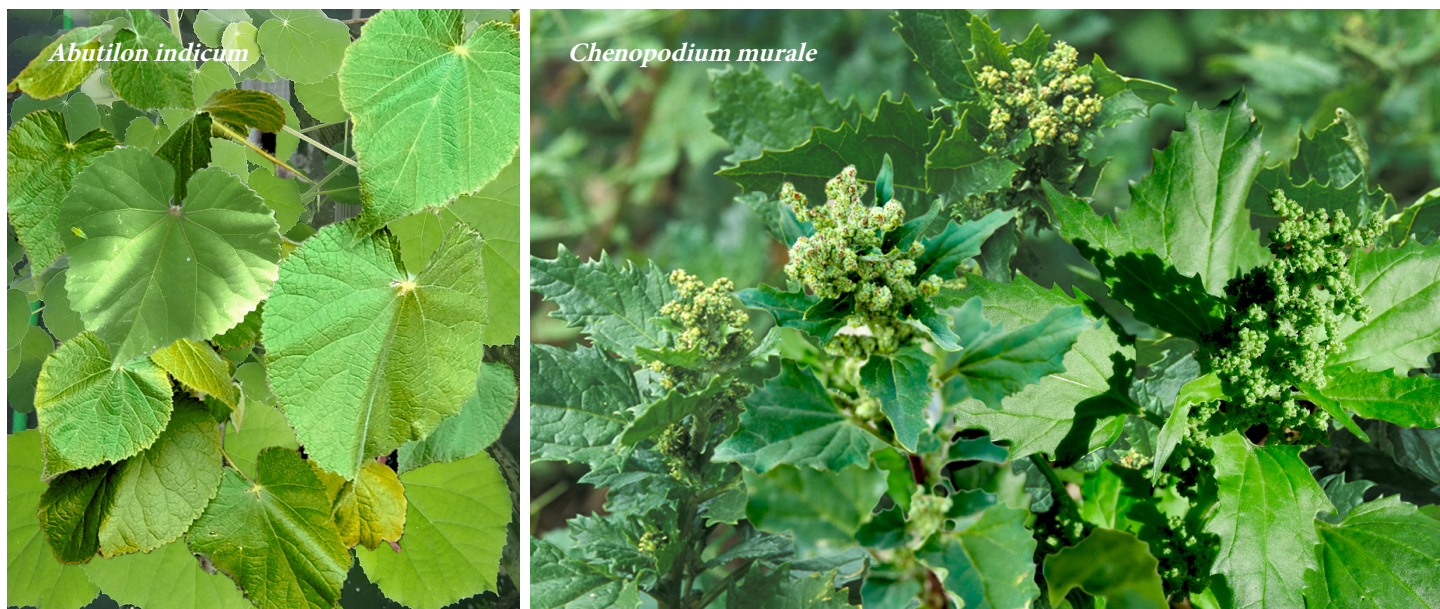


Figure 4. *Amrasca biguttula*'s survival on key weed hosts, such as *Abutilon indicum* (left) and *Chenopodium murale* (right), which sustain jassid populations adjacent to primary crops like cotton, sunflower, potato, cowpea, eggplant and okra. These weeds enable the pest to feed and proliferate year-round, bridging off-season gaps and facilitating continuous ecosystem persistence, even as main crops mature or are harvested. Moreover, jassids on these hosts serve as reservoirs for naturally occurring biological control agents, including predators and parasitoids, which can migrate to infested crops, potentially amplifying suppression during peak infestation periods.

Table-3. First Records of *Amrasca biguttula*

Year	Location	Region	Host Crops	Impact/Notes	Citation
2012	Zarrindasht, Fars Province, Iran	Middle East	Cotton	First Middle Eastern record; establishment on cotton fields	Taghizadeh <i>et al.</i> 2012
2017	Baghdad, Iraq	Middle East	Okra, egg-plant, pepper, cowpea, mallow	New record; serious pest of okra, population peaks in June and October, 30–155 leafhoppers/plant on older crops	Al-Hamadany & Al-Karboli, 2017
1999	Ghana	West Africa	Okra	First confirmed African invasion; outbreak on okra, escalating from low-level presence	Obeng-Ofori & Sackey, 2003; EPPO, 2024
2022	Côte d'Ivoire	West Africa	Cotton, okra, eggplant	Displaced <i>Jacobiasca lybica</i> (90–100% of jassid populations); up to 25% yield loss in cotton; exceeded economic thresholds	Kouadio <i>et al.</i> , 2024; PR-PICA, 2022
2022	Northern Cameroon	West Africa	Cotton	First record; 22–84% of leaf-hopper diversity, severe damage in 2023, peak in August	Jacques <i>et al.</i> 2024
2022–2023	Burkina Faso, Chad, Mali, Nigeria, Senegal, Togo, Benin	West Africa	Okra, egg-plant, cotton	Reported via technical/media sources; 50–100% losses in untreated fields	Dumbardon-Martial & Pierre, 2025; EPPO, 2024; Silvie <i>et al.</i> , 2023
2023 (April)	Southern Puerto Rico (Juana Díaz, Río Cañas Abajo)	Caribbean	Cotton (cultivated/wild), egg-plant	First Western Hemisphere record; raised concerns for spread to southeastern U.S. and Caribbean	Cabrera-Asencio <i>et al.</i> , 2023; USDA, 2025
2023 (Sept)	U.S. Virgin Islands (St. Croix)	Caribbean	Okra	Confirmed on okra; rapid regional spread	Dmitriev <i>et al.</i> , 2024; IPPC, 2024
2024	Antigua, Anguilla, St. Kitts and Nevis	Caribbean	Unspecified hosts	Confirmed detections; part of Caribbean expansion	EPPO, 2024; IPPC, 2024
2024	Niger (Niamey)	West Africa	Okra, guinea sorrel	New invasive species; 50–100% crop losses, confirmed morphologically	Akonde <i>et al.</i> , 2024
2024 (Feb)	Barbados	Caribbean	Okra, sorrel, cotton	Present, not widely distributed; second Caribbean record	EPPO, 2024; IPPC, 2024
2024 (Jan)	Martinique (Fort-de-France), France	Caribbean	Cotton	Detected on cotton; likely established for years	Dumbardon-Martial & Pierre, 2025
2024 (Nov-Dec)	Florida (Miami-Dade County), USA	North America	Okra	First U.S. continental record; spread to nurseries via hibiscus shipments	CAPS, 2025; Esquivel <i>et al.</i> , 2025
2025	Egypt (Kafr El Sheikh governorate)	North Africa	Cotton, roselle, okra	First North African record	El-Hady & El-Hashash, 2025
2025 (July)	Georgia Alabama South Carolina & southern counties of USA	North America	Cotton, okra	Rapid spread to 101 counties; severe hopperburn, >100 insects/leaf on okra; COI barcoding confirmed in Alabama (>99% identity)	Esquivel <i>et al.</i> , 2025; Zhao & Balkcom, 2025; Harper, 2025; NC State Extension, 2025

Within its native distribution, *A. biguttula* is widespread in warmer subtropical and tropical agroecosystems, attacking a broad array of economically important crops, including cultivated and wild cotton (*Gossypium* spp.), cultivated cotton species (*Gossypium hirsutum* and *G. barbadense*), eggplant (*Solanum melongena*), potato (*Solanum tuberosum*), okra (*Abelmoschus esculentus*), cowpea (*Vigna unguiculata*),

maize (*Zea mays*), sunflower (*Helianthus annuus*), soybean (*Glycine max*), peanut (*Arachis hypogaea*), mung bean (*Vigna radiata*), pigeon pea (*Cajanus cajan*), jute (*Corchorus* spp.), sorghum (*Sorghum bicolor*), ornamental hibiscus (*Hibiscus rosa-sinensis*) and niger (*Guizotia abyssinica*), which suffer economically significant hopperburn damage, characterized by yellowing, curling, and necrosis, leading to yield

losses of up to 40–100% in untreated fields (Al-Hamdany & Al-Karboli, 2017; CABI, 2024; Fatima *et al.*, 2021; Kamble & Sathe, 2015; Sahito *et al.*, 2017; Obeng-Ofori & Sackey, 2003). Among these, cotton, okra, eggplant, and sunflower are the most frequently studied due to their commercial importance and high susceptibility, with *A. biguttula* populations peaking during warm, dry conditions in May–June in regions like Southern Punjab, coinciding with cotton seedling emergence and leading to severe infestations (Ghani, 1946; Naveed, 2006; Saeed *et al.*, 2015).

Alternative hosts, both cultivated and wild, play a critical role in sustaining *A. biguttula* populations. A 2009 study identified 48 plant species across 22 taxonomic families as hosts for *A. biguttula*, with 24 classified as “true” hosts supporting both nymphs and adults, and 24 as “incidental” hosts harboring only adults temporarily (Baig *et al.*, 2009). True hosts, particularly in Malvaceae (*Abelmoschus esculentus*, *Hibiscus sabdariffa*, *Hibiscus rosa-sinensis*) and Euphorbiaceae (*Ricinus communis*), support high pest densities due to favorable chemical properties like crude protein and nitrogen content (Ghani, 1946; Iqbal *et al.*, 2011).

Okra, often intercropped with cotton, and eggplant, grown in close proximity, are primary reservoirs, sustaining year-round populations except in January–February due to adverse weather or plant maturity (Anitha, 2007; Eijaz *et al.*, 2012). Castor, *Ricinus communis*, a perennial grown on

marginal lands, serves as a continuous host with minimal insecticide exposure, maintaining pest populations during cotton off-seasons (Hattam & Abbasi, 1994; Yousafi *et al.*, 2013). Other notable true hosts include mallow (*Malva parviflora*), pepper (*Capsicum annuum*), and weeds like *Abutilon indicum* and *Chenopodium murale*, which harbor lower populations but act as refuges during inter-harvest periods, contributing to pest carry-over to cotton (Baig *et al.*, 2009; Huque, 1994; Srinivasan, 2009).

Additional hosts include maize (*Zea mays*), millet (*Sorghum* spp.), Bermuda grass (*Cynodon dactylon*), and grapevine (*Vitis vinifera*), though grapes are considered a minor host due to limited evidence of significant damage (Ghosh & Karmakar, 2021; Kulkarni, 2020). Ornamental hosts like *Hibiscus rosa-sinensis* show variable susceptibility, with some cultivars lightly colonized in Puerto Rico, while others suffer extensive damage (Nieves, 2024, personal communication). Seasonal host shifts occur as *A. biguttula* migrates to exploit available plants, with population densities peaking in warmer months and influenced by host growth stage, weather, and insecticide use (Barman *et al.*, 2010; Setamou *et al.*, 2000).

The diversity of hosts underscores the pest’s adaptability, complicating management, as weeds and alternative crops near cotton fields can sustain populations, necessitating integrated strategies targeting both primary and reservoir hosts to disrupt pest cycles.

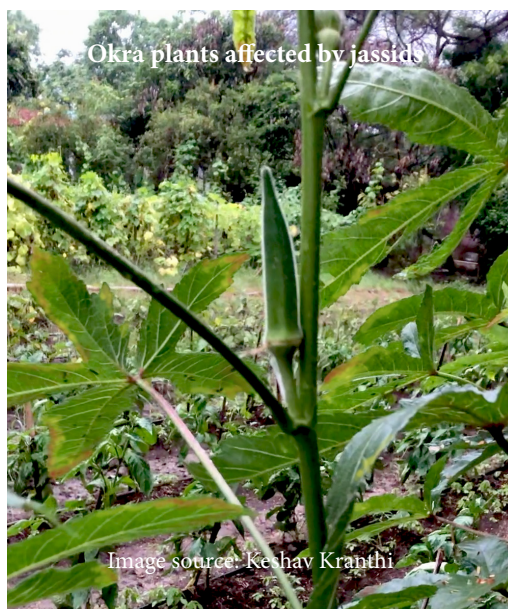


Figure-5. Leaf damage and hopper-burn Symptoms on Okra and Sunflower Infested by *Amrasca biguttula*

Impact of Host Crop Diversity on Natural Control of *Amrasca biguttula*

The polyphagous nature of *Amrasca biguttula*, allows it to exploit a wide range of true alternative host plants, which serve as critical reservoirs during the inter-harvest period when primary crops like cotton (*Gossypium hirsutum*) are absent (Baig *et al.*, 2009; Huque, 1994). These alternative hosts, spanning families such as Malvaceae (*Abelmoschus esculentus*, *Hibiscus sabdariffa*), Euphorbiaceae (*Ricinus communis*), and Solanaceae (*Solanum melongena*), sustain pest populations year-round, particularly in subtropical and tropical agroecosystems, facilitating migration to cotton crops and exacerbating infestations (Clementine *et al.*, 2005; Saeed *et al.*, 2015). However, these hosts also harbor natural enemies, including predators and para-

sitoids, which can mitigate pest pressure, creating a complex balance of ecological benefits and challenges that varies by host plant characteristics, such as growth habit, perenniality, and abundance (Sattar, 2010; Tscharrntke, 2000).

True alternative hosts like okra (*A. esculentus*), castor (*R. communis*), and eggplant (*S. melongena*) support both nymphs and adults of *A. biguttula*, acting as primary carry-over sources to cotton, especially during peak population periods in May–June (Baig *et al.*, 2009; Naveed, 2006). Weeds (*Abutilon indicum*, *Chenopodium murale*) and ornamentals (*Hibiscus rosa-sinensis*) further sustain low-density populations during off-seasons, amplifying pest persistence (Huque, 1994; Srinivasan, 2009). Conversely, these hosts foster natural enemy populations, offering potential for biological control. For example, *R. communis*, a perennial with minimal pesticide exposure, supports high densities of predators like *Chrysoperla carnea*, which feed on its pollen, and serves as a refuge due to its bushy canopy (Sattar, 2010). Vegetable hosts like okra and cucurbits (*Cucumis melo* var. *phutt*, *Luffa aegyptiaca*) also harbor predators and two key egg parasitoids, *Anagrus* sp. and *Arescon enocki*, which target *A. biguttula* eggs before feeding damage occurs (Sahito *et al.*, 2010; Wajnberg & Hassan, 1994).

The efficacy of natural enemies varies across hosts and agroecosystems. Parasitism rates by *A. enocki* (predominant on okra and castor) and *Anagrus* sp. (exclusive on *L. aegyptiaca*) reach 38.6%, slightly above the 32–36% threshold for effective biological control, but are inconsistent across other hosts, limiting their standalone impact (Tscharrntke, 2000; Sattar, 2010).

Factors influencing natural enemy success include plant volatile profiles, nectar availability, and morphology, which affect parasitoid attraction and competitive interactions (Hawkins, 2000; Jervis & Heimpel, 2005; Micha *et al.*, 2000). Additionally, intra-guild predation and pesticide susceptibility reduce natural enemy efficacy, particularly on heavily sprayed crops like okra and eggplant (Rosenheim *et al.*, 1995; Tscharrntke, 2000). Seasonal availability of hosts further complicates dynamics, with weeds and ornamentals providing year-round refuges, while crops like cotton and vegetables peak in availability during warmer months (Sattar, 2010).

The dual role of alternative hosts as pest reservoirs and natural enemy habitats creates a nuanced management challenge. While hosts like castor and okra are agriculturally beneficial, their role in sustaining *A. biguttula* can outweigh benefits in cotton agroecosystems unless natural enemy migration to crops is enhanced (Tscharrntke, 2000).

Diversifying agroecosystems by integrating non-host plants that support generalist predators, alongside targeted conservation biological control, could suppress *A. biguttula* populations, but requires further research into host-specific enemy interactions and pesticide impacts to optimize natural control across seasons (Naveed *et al.*, 2007).

Mode of Dispersal

Amrasca biguttula, exhibits a multifaceted dispersal strategy that combines limited active flight with passive mechanisms, enabling both short-range local spread and long-distance invasions (Blackmer *et al.*, 2004; Ghauri, 1982). Adults possess a short active movement capacity, typically limited to short flights or jumps when disturbed, with dispersal distances comparable to other leafhoppers like *Homalodisca* spp. (10–100 m), but this alone is insufficient for extensive range expansion (Northfield *et al.*, 2009). Instead, *A. biguttula* relies heavily on passive transport via wind currents, which can carry flying adults over long distances, as documented in historical cases of leafhopper dispersal spanning hundreds of kilometers (Ghauri, 1982). In the Caribbean, wind-assisted dispersal is implicated in the pest's rapid movement from Puerto Rico (2023) to neighboring islands like Barbados and Martinique (2024), and potentially to the continental U.S., where prevailing trade winds facilitate cross-regional transport (Andraca-Gómez *et al.*, 2020; Bertone *et al.*, 2008; Dumbardon-Martial & Pierre, 2025).

Human-mediated pathways represent the primary vector for international introductions, with *A. biguttula* hitchhiking on infested propagation materials, such as cotton seeds, bolls, or ornamental plants like hibiscus (*Hibiscus rosa-sinensis*), where eggs or nymphs adhere to foliage (ARM, 2024; CAPS, 2025). Agricultural inspectors have intercepted the pest five times in aircraft cargo at U.S. ports of entry, highlighting trade as an open but infrequent pathway, often via contaminated shipments from Asia, Africa, or the Caribbean (ARM, 2024). Eggs laid in soft plant tissues and nymphs on foliage enable survival during transport, while adults may disperse as stowaways on equipment, vehicles, or human clothing (Esquivel *et al.*, 2025; Zhao & Balkcom, 2025). In the southeastern U.S., rapid spread from Florida (November 2024) to 101 counties in Alabama, Georgia, and South Carolina by July 2025 likely involved a combination of wind and contaminated nursery stock, with infested hibiscus detected in retail outlets in Florida, Louisiana, and Texas (CAPS, 2025; NC State Extension, 2025).

The exact pathways for *A. biguttula*'s recent invasions remain unlinked to specific vectors, but its polyphagous habits and adaptability amplify risks through unregulated trade in host commodities (Cabrera-Asencio *et al.*, 2023; Kouadio *et al.*, 2024). To mitigate introductions, U.S. Plant Protection and Quarantine (PPQ) regulations under 7 CFR parts 305 and 319 outline phytosanitary measures for high-risk hosts, including inspections, phytosanitary certificates, post-entry quarantines, and mandatory treatments like fumigation or hot water dips for cotton seeds, okra, eggplant, and hibiscus from infested regions (APHIS, 2025a; APHIS, 2025b). The Phytosanitary Export Database and Agricultural Commodity Import Requirements (ACIR) database detail country-specific requirements, prohibiting untreated propagation materials from Asia and Africa and mandating certification that consignments are free of pests (APHIS, 2025c). Emerging modeling suggests that under climate change, wind-driven dispersal could accelerate range expansion in cotton belts, necessitating enhanced surveillance of trade pathways and integration of predictive tools for early detection (Azrag *et al.*, 2025).

Life Cycle

Amrasca biguttula, exhibits a hemimetabolous life cycle with egg, nymph, and adult stages, characterized by rapid development, high fecundity, and year-round reproduction in warm climates with continuous host availability (AVRDC, 2003; Ghosh & Karmakar, 2022).

The lifecycle duration varies with environmental conditions like temperature and humidity, ranging from 15 to 46 days, supporting up to 11 overlapping generations annually in regions like India (Mensah, 2006; Saeed *et al.*, 2015). Below, the life cycle stages are detailed based on studies primarily conducted on okra (*Abelmoschus esculentus*) and cotton (*Gossypium hirsutum*), with consistent findings across multiple sources.

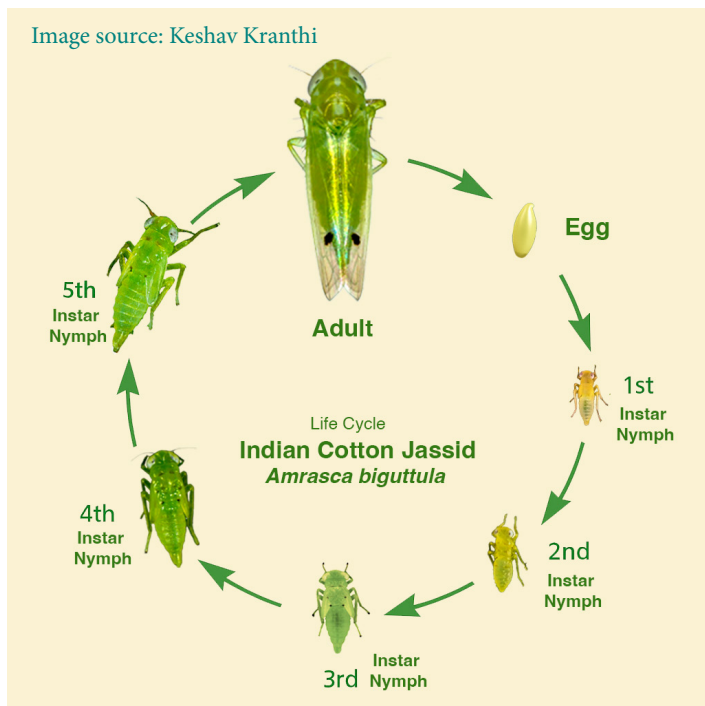


Figure-6. Life cycle of *Amrasca biguttula*

Eggs

Female *A. biguttula* insert yellowish-white, curved, oblong eggs (0.73×0.24 mm) into the spongy parenchymatous layer of leaf veins, midribs, petioles, or tender twigs, rendering them nearly invisible without magnification (Agarwal *et al.*, 1978; Mensah, 2006; Schreiner, 2000; Singh *et al.*, 2018). The incubation period averages 5.06 ± 1.16 days (range: 3–7 days) on okra, with similar durations on cotton (6.53 ± 0.58 days) and other hosts (6–10 days) (Jayasimha *et al.*, 2012; Shivanna *et al.*, 2009; Singh *et al.*, 2018).

Fecundity varies from 13–60 eggs per female, with averages reported as 15.66 ± 1.71 (Singh *et al.*, 2018), 17.20 (Thirumalaraju, 1984), 20.33 ± 2.65 (Shivanna *et al.*, 2009), or 25–34 eggs (Nagrare *et al.*, 2012), depending on host and conditions.



Figure-7. A schematic (artistic) image of *Amrasca biguttula* egg

Nymphs

Upon hatching, nymphs pass through five instars, progressing from transparent, creamy-whitish forms to yellowish-green, with a total nymphal period of 8.04–11.68 days (Jayasimha *et al.*, 2012; Shivanna *et al.*, 2009). Nymphs are flattened, pale yellowish-green, and exhibit characteristic diagonal or sideways movement when disturbed (Schreiner, 2000; Singh *et al.*, 2018). Developmental durations on okra include:

First instar: 1.49 ± 0.33 days (range: 1–3 days), creamy-whitish with blackish-brown eyes and setaceous antennae (Jayasimha *et al.*, 2012; Singh *et al.*, 2018).

Second instar: 1.19 ± 0.22 days (range: 2–5 days), with dull rudimentary wing pads (Jayasimha *et al.*, 2012; Singh *et al.*, 2018).

Third instar: 1.16 ± 0.15 days (range: 2–6 days), yellowish-green with prominent wing pads (Jayasimha *et al.*, 2012; Singh *et al.*, 2018).

Fourth instar: 1.60 ± 0.39 days (range: 3–6 days), with wing pads extending to the fourth abdominal segment (Jayasimha *et al.*, 2012; Singh *et al.*, 2018).

Fifth instar: 1.97 ± 0.29 days (range: 3–6 days), with wing pads reaching the ninth abdominal segment (Jayasimha *et al.*, 2012; Singh *et al.*, 2018).



Figure-8. First instar nymphal stage of *Amrasca biguttula*

Total nymphal duration averages 8.04 ± 0.51 days (Jayarao *et al.*, 2015) to 11.68 ± 3.74 days (Shivanna *et al.*, 2009), with shorter durations in warmer conditions.

Figure-9. Different nymphal stages of *Amrasca biguttula*

Adults

Adults are pale green, elongate, wedge-shaped, approximately 2–3.5 mm long, with yellowish-green to reddish-brown forewings (season-dependent) and diagnostic black spots: two on the vertex of the head (sometimes faint) and one near the apex of each forewing's brachial cell (Cabrera-Asencio *et al.*, 2023; Schreiner, 2000).

Adult jassids are highly active, displaying rapid sideways hopping or short flights when disturbed (Ghosh & Karmakar, 2022). Male longevity averages 15.80 ± 1.61 days (range: 12–19 days), while females live slightly longer at 16.38 ± 1.83 days (range: 12–22 days), with some studies reporting up to 34 days for males and 37 days for females (Nagrare *et al.*, 2012; Singh *et al.*, 2018).

The preoviposition period is 3.30 ± 0.35 days, oviposition lasts 4–9 days (mean: 9.53 ± 1.11 days), and post-oviposition is 3.80 ± 0.63 days (Shivanna *et al.*, 2009). The entire lifespan ranges from 19–35 days (mean: 27.63 ± 4.42 days), enabling multiple generations in tropical climates (Nagrare *et al.*, 2012).

**Figure-10.** *Amrasca biguttula* adult

Reproductive Dynamics

In warmer climates with continuous host availability (e.g., okra, cotton, hibiscus), *A. biguttula* breeds year-round, producing up to 11 generations annually due to overlapping reproductive cycles (AVRDC, 2003; Ghosh & Karmakar, 2022; Mensah, 2006).

In India, generation times range from 15–46 days, influenced by temperature, humidity, and host quality, with faster cycles in summer (Saeed *et al.*, 2015). This reproductive flexibility, coupled with high fecundity and short development times, enhances the pest's invasive potential, necessitating targeted monitoring during peak egg-laying periods (April–June) to disrupt population buildup in invaded regions like the southeastern U.S. and Caribbean (Esquivel *et al.*, 2025).

Table-4. Characteristics of different life stages of *Amrasca biguttula* on okra and cotton

Stage	Duration (Days)	Characteristics	Notes	Citation
Egg	Mean: 5.06 ± 1.16 (range: 3–7); 6.53 ± 0.58 on cotton; 6.42 ± 0.37 or 6.55 ± 0.40 on okra	Yellowish-white, curved, oblong (0.73×0.24 mm), inserted into leaf veins/midribs, petioles, or twigs in parenchymatous layer	Nearly invisible without magnification; fecundity 13–60 eggs/female (mean: 15.66 ± 1.71 to 20.33 ± 2.65)	Agarwal <i>et al.</i> , 1978; Jayasimha <i>et al.</i> , 2012; Mensah, 2006; Schreiner, 2000; Shivanna <i>et al.</i> , 2009; Singh <i>et al.</i> , 2018; Thirumalaraju, 1984
First Instar Nymph	Mean: 1.49 ± 0.33 to 2.06 ± 0.79 (range: 1–3)	Creamy-whitish, transparent, rounded head, blackish-brown eyes, setaceous antennae, no wings	Active, diagonal movement when disturbed	Jayasimha <i>et al.</i> , 2012; Shivanna <i>et al.</i> , 2009; Singh <i>et al.</i> , 2018
Second Instar Nymph	Mean: 1.19 ± 0.22 to 3.53 ± 1.06 (range: 2–5)	Dull rudimentary wing pads on meso- and metathorax, longer setaceous antennae, 1.05–1.10 mm long	Yellowish-green, mobile	Jayasimha <i>et al.</i> , 2012; Singh <i>et al.</i> , 2018
Third Instar Nymph	Mean: 1.16 ± 0.15 to 3.67 ± 1.11 (range: 2–6)	Yellowish-green, prominent wing pads	Increasingly active, side-ways movement	Jayasimha <i>et al.</i> , 2012; Singh <i>et al.</i> , 2018
Fourth Instar Nymph	Mean: 1.60 ± 0.39 to 4.40 ± 1.12 (range: 3–6)	Yellowish-green, wing pads to fourth abdominal segment, active	Distinct wing pad development	Jayasimha <i>et al.</i> , 2012; Singh <i>et al.</i> , 2018
Fifth Instar Nymph	Mean: 1.97 ± 0.29 to 4.73 ± 1.09 (range: 3–6)	Greenish-yellow, wing pads to ninth abdominal segment, long setaceous antennae	Highly mobile, pre-adult stage	Jayasimha <i>et al.</i> , 2012; Singh <i>et al.</i> , 2018
Total Nymphal Period	Mean: 8.04 ± 0.51 to 11.68 ± 3.74 (range: 5–16)	Five instars, pale yellowish-green, flattened, diagonal movement	Duration varies with temperature, shorter in warmer conditions	Jayarao <i>et al.</i> , 2015; Nagrare <i>et al.</i> , 2012; Shivanna <i>et al.</i> , 2009
Adult	Male: 15.80 ± 1.61 to 15.90 ± 1.58 (range: 12–34); Female: 16.38 ± 1.83 to 18.66 ± 1.88 (range: 12–37)	Pale green, 2–3.5 mm, wedge-shaped, yellowish-green to reddish-brown wings (seasonal), black spots on head (2, sometimes faint) and forewings (1 per wing)	Preoviposition: 3.30 ± 0.35 days; oviposition: 4–9 days (mean: 9.53 ± 1.11); post-oviposition: 3.80 ± 0.63 days; rapid hopping/flight when disturbed	Cabrera-Asencio <i>et al.</i> , 2023; Nagrare <i>et al.</i> , 2012; Schreiner, 2000; Shivanna <i>et al.</i> , 2009; Singh <i>et al.</i> , 2018
Total Lifespan	Mean: 27.63 ± 4.42 (range: 19–35)	Egg to adult, influenced by climate and host quality	Up to 11 generations/year in warm climates with continuous hosts	Ghosh & Karmakar, 2022; Mensah, 2006; Nagrare <i>et al.</i> , 2012; Saeed <i>et al.</i> , 2015

Impact of Sowing Date on Pestilence

The timing of cotton sowing significantly influences the population dynamics and pestilence of *Amrasca biguttula*, due to its interaction with environmental factors and host plant phenology (Saeed *et al.*, 2018; Sunita Devi *et al.*, 2019). Early sowing (March–April) generally reduces pest pressure and insecticide requirements, while late sowing (May–August) increases vulnerability to *A. biguttula* infestations, particularly as crops mature or alternative hosts become scarce, necessitating strategic planting schedules within integrated pest management (IPM) frameworks (Esquivel *et al.*, 2025; Saeed *et al.*, 2018).

Field experiments conducted during the kharif seasons of 2013 and 2014 in India demonstrated that sowing date significantly affects leafhopper populations on cotton under unsprayed conditions (Sunita Devi *et al.*, 2019). Early sowing resulted in a lower average population of 5.60 leafhoppers per plant, compared to 6.03 leafhoppers per plant in late-

sown crops. Peak infestations occurred at the 38th standard week (mid-September), with early-sown crops recording 11.50–13.73 leafhoppers per plant and late-sown crops reaching 16.10–20.40 leafhoppers per plant, indicating higher pest pressure in later plantings (Sunita Devi *et al.*, 2019). Weather parameters further modulated these dynamics: in early-sown crops, leafhopper populations showed a positive correlation with temperature, relative humidity, and rainfall, but a negative correlation with sunshine hours, while in late-sown crops, minimum temperature and wind speed were positively correlated (Sunita Devi *et al.*, 2019).

Studies on action thresholds (ATs) for *A. biguttula* management in conventional (CIM-554) and transgenic (Bt.CIM-599) cotton cultivars across three planting dates (15 March, 15 April, 15 May) in 2011–2012 further highlight the impact of sowing time (Saeed *et al.*, 2018). Cotton planted on 15 March (early sown) exhibited the lowest pest pressure, with

populations never exceeding 2 leafhoppers per leaf, requiring only one insecticidal application at an AT of 1.0 leafhopper per leaf, yielding the highest seed cotton yield (up to 3,500 kg/ha), net return, and marginal rate of return (MRR). In contrast, the regular planting date, 15 April plantings required 3 applications at an AT of 2.0 leafhoppers per leaf (vs. 10 at 0.1 AT) to maintain comparable yields, while 15 May (late plantings) were most vulnerable, requiring 4 applications at an AT of 1.0 leafhopper per leaf to optimize returns (Saeed *et al.*, 2018).

Late-planted fields faced heightened risk as *A. biguttula* migrated from defoliated or harvested older cotton fields, particularly post-defoliation in late summer, exacerbating infestations in younger, late-sown crops (Esquivel *et al.*, 2025).

These findings indicate that early sowing minimizes *A. biguttula* pestilence by aligning crop growth with less favorable conditions for population buildup and reducing reliance on insecticides, enhancing cost-effectiveness within IPM programs (Saeed *et al.*, 2018).

Impact of Fertilizers on Pestilence

The application of fertilizers, particularly nitrogen (N), phosphorus (P), and potassium (K), significantly influences the population dynamics and pestilence of *Amrasca biguttula* on crops such as eggplant (*Solanum melongena*) and cotton (*Gossypium hirsutum*), with higher nitrogen levels generally exacerbating infestations (Godase & Patel, 2001; Sagar *et al.*, 2014). The nutritional status of host plants, altered by fertilizer regimes, affects pest development rates, population density, and crop susceptibility, necessitating balanced fertilization strategies within integrated pest management (IPM) programs to minimize *A. biguttula* damage (Esquivel *et al.*, 2025).



Figure-11. Bushy vegetative growth of cotton due to application of excessive nitrogenous fertilizers.

Field experiments conducted at Navsari, Gujarat, India, evaluated the impact of nine fertilizer treatments on eggplant, including organic manures (farmyard manure [FYM], vermicompost, neem cake) and inorganic NPK combinations, on *A. biguttula* populations (Godase & Patel, 2001). The highest jassid population (24.71 per 9 leaves) was recorded with a double nitrogen dose (200:37.5:37.5 kg NPK/ha, T6), where nymphs developed fastest (9.86 days, growth index 7.61), indicating a preference for nitrogen-rich plants.

In contrast, a double potassium dose (50:37.5:75 kg NPK/ha, T7) resulted in the lowest jassid population (9.14 per 9 leaves) and slowest nymphal development (11.41 days, growth index 5.26). Neem cake alone (1.7 t/ha, T5) also reduced pest populations, suggesting that potassium and neem cake has a negative effect on jassid populations and higher nitrogen doses favor jassid infestation (Godase & Patel, 2001).

Nitrogen contents and crude protein were found to have significant and positively correlated with jassid population infestation with nitrogen having the maximum impact (Khan *et al.*, 2017). Similarly, field experiments on cotton (Sagar *et al.* 2014) with nitrogen dosages (0–200 kg/ha) showed a significant positive correlation between nitrogen levels and *A. biguttula* populations, with the highest leafhopper densities at 150–200 kg/ha compared to recommended (100 kg/ha) or lower doses.

Laboratory bioassays corroborated these findings, showing enhanced pest reproduction and survival on nitrogen-enriched plants (Sagar *et al.*, 2013c). Field observations further indicate that potassium-deficient cotton fields exhibit greater susceptibility to *A. biguttula*, with weaker plants showing pronounced hopper-burn symptoms (e.g., chlorosis, necrosis), suggesting potassium's role in enhancing plant resistance.

These results highlight that excessive nitrogen fertilization promotes *A. biguttula* pestilence by enhancing host plant suitability, while potassium and organic amendments like neem cake can mitigate infestations. Thus, optimizing fertilizer regimes—balancing nitrogen reduction with potassium supplementation—could reduce pest pressure and support sustainable IPM, particularly under climate-driven increases in pest activity.

Impact of Crop Density on Pestilence

Studies (Priyanka *et al.*, 2017; Rajasekhar *et al.*, 2018) showed that planting density significantly influences the pestilence of *Amrasca biguttula* in cotton (*Gossypium hirsutum*) agroecosystems, with higher densities planting systems (HDPS) exacerbating leafhopper populations compared to normal spacing. Increased plant density creates favorable microclimates and resource availability, promoting pest buildup, which necessitates targeted management strategies within integrated pest management (IPM) frameworks to mitigate economic damage (Rajasekhar *et al.*, 2018).



Figure-12 A high density crop

Field experiments conducted during 2015–2016 at Lam, Guntur, Andhra Pradesh, India, showed that leafhopper populations ranged from 1.60 to 18.27 per three leaves across crop growth stages, with the lowest density (1.60 leafhoppers per three leaves) recorded at 93 days after sowing (DAS) in low-density plots (14,814 plants per hectare). In contrast, the highest infestation (18.27 leafhoppers per three leaves) occurred at 45 DAS in HDPS plots (111,111 plants per hectare). The overall mean population increased from 5.03 to 6.97 leafhoppers per three leaves as plant density rose from 14,814 to 111,111 plants per hectare, demonstrating a positive correlation between planting density and leafhopper incidence (Priyanka *et al.*, 2017).

Similar findings were reported from a 2016–2017 study at the same location, comparing *A. biguttula* populations under HDPS and normal spacing in rainfed cotton (Rajasekhar *et al.*, 2018). In HDPS, leafhopper populations exceeded the economic threshold level (ETL) of 6 leafhoppers per three leaves at 45 and 75 DAS (both 6.25 per three leaves), while normal spacing maintained populations below the ETL throughout the season. Among tested insecticides, flonicamid 50% WP at 0.3 g per litre effectively reduced leafhopper populations in both systems, highlighting the need for chemical interventions in high-density settings (Rajasekhar *et al.*, 2018). The increased pest pressure in HDPS is likely due to denser canopies enhancing humidity and reducing natural enemy efficacy, creating ideal conditions for *A. biguttula* proliferation (Priyanka *et al.*, 2017). These results indicate that HDPS, while potentially increasing yield potential, heightens *A. biguttula* pestilence, requiring more intensive monitoring and control measures compared to normal spacing.

Impact of Weather

The population dynamics of the *Amrasca biguttula* are governed by a complex interplay of abiotic factors, with temperature, humidity, and rainfall emerging as the most influential drivers. A consistent pattern across multiple studies is the seasonal peak of leafhopper populations during the hottest months of the year, typically from June to August in regions like South Asia (Ahmad *et al.*, 1985; Ghosh and Karmakar,

2022; Jayasimha *et al.*, 2012; Mensah, 2006; Saeed *et al.*, 2015; Thirasack, 2001). Nymphs and adults predominantly inhabit the abaxial side of leaves, and their population peaks are consistently associated with a specific range of environmental conditions, notably temperatures between 29–35°C (81°F and 93°F) and relative humidity around 70% (Ghosh and Karmakar, 2021; Mohammad *et al.*, 2019; Rajasekhar *et al.*, 2015).

The relationship between temperature and leafhopper incidence, however, is nuanced and requires differentiation between maximum and minimum temperatures. Multiple studies report a significant positive correlation between leafhopper populations and minimum temperature (Devi *et al.*, 2019; Janu *et al.*, 2017; Nemade *et al.*, 2015; Sharma and Sharma, 1997; Srinivasan *et al.*, 1986). This is likely because higher minimum temperatures facilitate faster development and reproductive rates, as lower temperatures have been shown to decrease growth rates (Sidhu and Dhawan, 1981; Srinivasan *et al.*, 1986). In contrast, the effect of maximum temperature is more variable. Several studies found a significant negative correlation, suggesting that excessively high temperatures may be inhibitory (Hussain *et al.*, 2014; Janu *et al.*, 2017; Sandhi and Sidhu, 2018; Sharma and Sharma, 1997). However, other research indicates a positive correlation with both maximum and minimum temperatures (Marbet *et al.*, 1984; Pati *et al.*, 2018; Rajasekhar *et al.*, 2015), highlighting that the optimal thermal range is broad, and the specific regional context, including the interplay with other factors, is critical.

Humidity is another critical factor, with evening relative humidity frequently showing a strong positive correlation with leafhopper population build-up (Devi *et al.*, 2019; Rajasekhar *et al.*, 2015; Nemade *et al.*, 2015; Sandhi and Sidhu, 2018). High humidity levels, particularly in the range of 55–87%, appear to create favourable conditions for survival and reproduction (Rajasekhar *et al.*, 2015). This is further supported by studies that found positive correlations with both morning and evening relative humidity (Nemade *et al.*, 2015; Janu *et al.*, 2017). Jassids population exhibited positive correlation with average temperature relative humidity, rainfall, rainy days and wind velocity (Shitole *et al.*, 2009). However, some studies report a negative association with morning humidity or find its influence to be non-significant when considered independently, underscoring the complex and sometimes contradictory role of this parameter (Pati *et al.*, 2018; Rajasekhar *et al.*, 2015).

Perhaps the most consistently documented abiotic mortality factor is rainfall. Heavy rainfall acts as a key mortality factor for both nymphs and adults, physically dislodging and killing them (Lal and Mahal, 1990). Periods of continuous rain, particularly when combined with temperatures lower than 29°C, humidity higher than 78%, and reduced sunshine, can drastically reduce pest populations by over 70% (Lal and Mahal, 1990). Consequently, population densities typically decrease during periods of high rainfall (Patel and Radadia,

2018; Ghosh and Karmakar, 2021; Mohammad *et al.*, 2019). Despite this clear negative mechanical impact, several correlation studies have reported positive relationships between rainfall and leafhopper incidence (Hussain *et al.*, 2014; Nemade *et al.*, 2015; Rajasekhar *et al.*, 2015; Sandhi and Sidhu, 2018). This apparent contradiction may be because moderate, non-damaging rainfall can increase humidity and promote succulent plant growth, indirectly favouring leafhoppers, whereas heavy, intense rainfall is directly detrimental.

Jassid population showed a significant positive correlation with sunshine hours and a significant negative correlation with wind speed and rainy days (Patel and Radadia, 2018). However, the complexity of these interactions is evident in multivariate analyses. For instance, Rajasekhar *et al.* (2015) found that weather variables collectively accounted for 57.2% of the variation in leafhopper populations, yet no single factor exerted a significant independent influence. Similarly, Sandhi and Sidhu (2018) noted that while individual parameters like sunshine hours and wind speed had low individual correlation coefficients, all parameters combined yielded a very high coefficient of determination ($R^2 = 0.96$), indicating that the population dynamics are a result of the synergistic effect of all ecological factors acting in concert. This holistic view is essential for effective forecasting, as the influence of one factor, such as temperature, can be mediated or even reversed by others like humidity and rainfall (Pathania, 2020). The determination of these combined effects is crucial not only for predicting outbreaks but also for formulating timely and effective integrated pest management strategies (Pati *et al.*, 2018).

Influence of Weather on Jassid Predators

The efficacy of natural enemies in regulating *Amrasca biguttula* populations is not constant but is significantly modulated by abiotic conditions, which differentially affect various predator species. Research indicates that key predators, including coccinellids (ladybird beetles), chrysopids (lacewings), and spiders, each exhibit distinct and sometimes contrasting responses to weather parameters, which in turn influences their temporal effectiveness in biological control.

Temperature exerts a species-specific influence on predator populations. Notably, minimum temperature shows a significant positive relationship with coccinellid abundance, suggesting that warmer nights are favourable for their population build-up (Nemade *et al.*, 2015). In contrast, ladybird beetle populations have been found to have a highly significant negative correlation with both minimum and average temperature, indicating that warmer conditions may be less favourable for this particular predator (Patel and Radadia, 2018). Maximum temperature has a varied impact on different predators; it is highly significantly and positively correlated with chrysopid and spider populations (Nemade *et al.*, 2015; Patel and Radadia, 2018).

Humidity and rainfall also play a critical role in shaping predator dynamics. Rainfall appears to be very favourable for coccinellids, showing a highly significant positive impact on their population build-up alongside jassids (Nemade *et al.*, 2015). Similarly, both morning and evening relative humidity are positively correlated with coccinellid numbers (Nemade *et al.*, 2015).

Conversely, other studies found that ladybird beetle populations declined with high humidity and rainfall, showing a highly significant negative correlation with these parameters (Patel and Radadia, 2018). Furthermore, chrysopids and spiders also exhibited significant negative correlations with rainy days and wind speed (Patel and Radadia, 2018). Solar radiation is another key factor, with bright sunshine hours being favourable for chrysopid egg development and positively correlated with spider and ladybird beetle activity (Nemade *et al.*, 2015; Patel and Radadia, 2018).

These differential responses to weather directly influence the temporal pest control efficacy of the predator guild. For instance, Nemade *et al.* (2015) observed that coccinellids, which thrive under the same conditions of high minimum temperature and humidity that benefit early-season jassids, show a significant positive relationship with the pest population in the early phase, demonstrating a strong numerical response. In the later phase, however, spiders, which are favoured by high maximum temperatures and sunshine, become more effective controllers of the leafhopper population.

The impact of chrysopids may be more limited, as some studies indicate a non-significant correlation with jassid population control despite their abundance being weather-dependent (Nemade *et al.*, 2015). Consequently, the biological control of jassids is a dynamic process where seasonal shifts in weather parameters can alter the dominance and effectiveness of different natural enemy species within the agro-ecosystem.

Impact of Naturally Occurring Biological Control on *Amrasca biguttula*

Naturally occurring biological control plays a significant role in suppressing populations of *Amrasca biguttula*, through a diverse suite of predators and parasitoids found on its host plants, particularly okra (*Abelmoschus esculentus*), cotton (*Gossypium hirsutum*), and alternative hosts like castor (*Ricinus communis*) (Adachi-Hogomori *et al.*, 2020; Wagan & Wagan, 2015).

These natural enemies, including spiders, predatory insects, and egg parasitoids, exploit the pest's eggs, nymphs, and adults, with their efficacy influenced by host plant characteristics such as type, abundance, and perennial nature, offering potential for integration into integrated pest management (IPM) strategies (Sahito *et al.*, 2010).



Image source: Keshav Kranthi

Figure-13 *Chrysoperla carnea* adult.

Image source: Keshav Kranthi

Figure-14 *Chrysoperla carnea* grub feeding on jassid nymph

Image source: Keshav Kranthi

Figure-15 Ladybird grub feeding on jassid nymph

Image source: Keshav Kranthi

Figure-16 Ladybird beetle adult.

Surveys in Pakistan on okra crops identified key predators, including spiders (Lycosidae, Thomisidae), lady beetles (*Coccinella septempunctata*, *C. undecimpunctata*, *Hyperaspis maindroni*, *Scymnus nubilus*, *Menochilus sexmaculatus*, *Brumus suturalis*), ants, green lacewings (*Chrysoperla carnea*), and hemipterans (*Orius* spp., likely *O. insidiosus*; *Geocoris* spp., likely *G. punctipes*), with spiders and coccinellids being the most abundant (Mari *et al.*, 2007; Wagan & Wagan, 2015). These predators were found on 23 true alternative host plants, with *R. communis* (Euphorbiaceae) harboring the highest density due to its perennial, bushy canopy, low pesticide exposure, and pollen availability for *C. carnea* (Sattar, 2010). Predators were three times more prevalent on crop plants than vegetables, with the lowest density on weeds, fruit plants, and ornamentals, except for *C. carnea* on rare plants and *Orius* spp. on fruit plants (Wagan & Wagan, 2015). Plant characteristics (e.g., abundance, shrub-like growth) signifi-



Image created by Keshav Kranthi

Figure-17 *Anagrus* wasp ovipositing in jassid egg (recreated artistic depiction based on *Anagrus* image from Jeong Yoo (<https://www.instagram.com/p/DDpXhV8p9fC/>))

cantly affected predator density, with perennial crops like *R. communis* providing optimal habitats (Sattar, 2010).

Egg parasitoids, notably *Anagrus empoascae* Dozier, *Stethynium empoascae* Subba Rao, *A. japonicus* and *Arescon enocki* (Hymenoptera: Mymaridae), target *A. biguttula* eggs embedded in leaf veins, offering early-stage control before feeding damage occurs (Adachi-Hogomori *et al.*, 2020; Rao *et al.*, 1968; Sahito *et al.*, 2010). In Okinawa, Japan, *A. japonicus* was identified as a novel egg parasitoid of *A. biguttula* on okra, with *A. enocki* emerging in smaller numbers (Adachi-Hogomori *et al.*, 2020).

Across four host plants (okra, *Cucumis melo* var. phutt, *Luffa aegyptiaca*, *R. communis*), parasitoids achieved an overall parasitism rate of 38.6% (± 0.03 SE), slightly above the 32–36% threshold for effective biological control (Tscharntke, 2000; Wagan & Wagan, 2015). Parasitoid distribution varied: *Anagrus* sp. dominated on *C. melo* var. phutt (100%) and *L. aegyptiaca* (83.3%), while *A. enocki* was more prevalent on okra (86.2%) and *R. communis* (Wagan & Wagan, 2015).

Parasitoid abundance was significantly influenced by plant abundance and perenniality but not growth habit, with no emergence from weeds, ornamentals, or fruit plants (Wagan & Wagan, 2015).

In regions like Puerto Rico, where *A. biguttula* was first reported in the Western Hemisphere in 2023, local predators such as *Chrysopodes collaris* (Neuroptera: Chrysopidae), *Orius insidiosus* (Hemiptera: Anthocoridae), *Zelus longipes* (Hemiptera: Reduviidae), *Chrysotus* spp. (Diptera: Dolichopodidae), *Taeniaptera* spp. (Diptera: Micropezidae), *Chilocorus cacti*, *Cycloneda sanguinea*, *Hippodamia convergens* (Coleoptera: Coccinellidae), *Solenopsis geminata* (Hymenoptera: Formicidae), *Polistes crinitus* (Hymenoptera: Vespidae), *Enallagma civile* (Odonata: Coenagrionidae), and miscellaneous spiders have been reported as predators of other Empoascini leafhoppers but lack confirmed predation on *A. biguttula* (Cabrera-Asencio *et al.*, 2023; Cotte & Cruz, 1989). This suggests potential gaps in local biological control efficacy, necessitating further research to evaluate the adaptability of these predators to *A. biguttula* in invaded regions (Cabrera-Asencio *et al.*, 2023).

Despite their potential, the efficacy of natural enemies is limited by factors such as intra-guild predation, pesticide susceptibility, and variable parasitism rates across hosts (Rosenheim *et al.*, 1995; Tscharntke, 2000).

In *A. biguttula*-invaded regions like the southeastern U.S., conserving natural enemy populations on alternative hosts through eco-friendly pesticide use and habitat diversification could enhance biological control, particularly during peak pest seasons. However, the lack of confirmed predation by local predators in regions like Puerto Rico highlights the need for studies on their behavioral adaptation to *A. biguttula* to optimize IPM strategies under climate-driven pest expansions

Jassid Surveillance and Scouting

Surveillance of *Amrasca biguttula* is a critical component of integrated pest management programs, as timely detection enables effective control before economic injury thresholds are surpassed. Confirmatory identification of jassids typically requires adult males; hence, collection of multiple individuals is recommended to ensure accurate diagnosis.

Surveillance activities are most effective when synchronized with the pest's population dynamics and prevailing climatic conditions. Visual surveys should be prioritized during warm, which favor higher jassid activity. In tropical and subtropical regions, populations remain active year-round, albeit at lower levels during cooler periods. Conversely, in temperate regions, surveys may be suspended during winter months when jassid activity ceases. End-of-season surveys are particularly valuable, as reduced pesticide applications increase the probability of detecting active populations.



Image source: Keshav Kranthi

Figure-18 A farmer scouting for jassids in a cotton field in India

A range of sampling approaches can be deployed depending on the survey objectives and field conditions. Trapping with yellow sticky cards placed 20–30 cm above host plants can capture adults but lacks species specificity, often collecting numerous non-target insects. Visual inspection and manual collection using aspirators, sweep nets, or beat methods remain the most reliable techniques for targeted surveillance. Nymphs and adults congregate on the undersides of host leaves, and their capture can be facilitated by tapping foliage over trays containing ethanol, isopropanol, or soapy water. Sweep net sampling, although efficient for quantitative assessments, should be employed cautiously in cotton and vegetable crops to minimize mechanical damage to flowers, bolls, or fruits. Similarly, beat sampling into alcohol-filled trays allows for effective recovery of live specimens for subsequent laboratory identification.

Survey site selection should prioritize fields where host crops such as cotton, eggplant, okra, and sunflower are grown, particularly those that are poorly managed or left unharvested, as such habitats can harbor large populations. Within each site, systematic sampling across representative field sections

is recommended; however, perimeter surveys may suffice in certain contexts, particularly where access is limited. Collected specimens should be preserved appropriately: live insects maintained in cool storage until processing, followed by freezing for 24 hours before sorting, and eventual preservation in 75–90% ethanol for identification. When expertise for taxonomic resolution is unavailable, specimens should be forwarded to trained identifiers to avoid misclassification with morphologically similar species.

The adoption of standardized surveillance protocols ensures consistency across regions and facilitates reliable monitoring of pest spread. Incorporating a combination of visual inspection, manual sampling, and sticky trap deployment within an appropriately designed survey framework enhances detection efficiency. These approaches provide the foundation for risk assessment, early warning systems, and implementation of timely management interventions against the cotton jassid.

Pest Scouting and Jassid Damage Grades



Figure-19 Jassid damage grades

Pest scouting is an essential component of integrated pest management for cotton jassid (*Amrasca biguttula*), as it provides timely information on population dynamics and crop damage.

Field observations indicate that jassids are often more concentrated on the third, fourth, or fifth mainstem leaves below the terminal, making these positions suitable for targeted scouting.

To standardize assessment, a leafhopper injury grading system has been developed to classify damage intensity. The grades are as follows:

Grade 1: Jassids present on plants, but leaves remain free from crinkling or yellowing at the margins.

Grade 2: A few leaves in the lower half of the canopy exhibit curling, crinkling, and slight yellowing along the leaf margins.

Grade 3: Widespread crinkling and curling of leaves all across the plant; yellowing, bronzing, and browning of leaves appear in the middle and lower canopy, with noticeable growth reduction.

Grade 4: Severe curling, yellowing, bronzing, browning, drying, and shedding of leaves, accompanied by pronounced stunting of plants.

The cumulative effect of these grades can be quantified through the Leafhopper Resistance Index (also referred to as the *Hopper-burn Index), proposed by Nageswara Rao (1973). The index is calculated using the formula:

$$\text{*Leafhopper resistance index or Hopper-burn Index} = \frac{(G_1 \times P_1) + (G_2 \times P_2) + (G_3 \times P_3) + (G_4 \times P_4)}{(P_1 + P_2 + P_3 + P_4)}$$

Explanation:

- G represents a score for the level of leafhopper damage or injury on a plant (likely on a scale, e.g., from 1 to 5).
- P represents the number of plants assigned that particular score.
- The subscripts 1, 2, 3, 4 refer to the different categories or grades on the damage scale.

The formula calculates a weighted average, where the damage score for each grade is multiplied by the number of plants exhibiting that grade.

These products are then summed and divided by the total number of plants observed. This provides a single, comprehensive value that reflects the overall leafhopper resistance of the plant variety being tested. A lower index value indicates higher resistance.

Based on this index, when an average of >2.0 nymphs or adults are present per leaf in 5 leaves examined per plant, in the plants included in sampling, the genotypes can be categorized for resistance as follows:

<1.0 : Highly resistant

1.0 – 2.0: Resistant

2.0 – 2.5: Intermediate

> 2.5 : Susceptible

This system provides a reliable framework for evaluating varietal resistance under field conditions, facilitating screening in breeding programs as well as guiding pest management decisions in farmers' fields.

The resistance index can also be directly translated into actionable economic thresholds for pest management. When sampling 40 random plants per acre, a biological or botanical control intervention is recommended at an index value of approximately 1.5. If the index escalates to a value between 1.5 and 2.0, the application of chemical control measures is advised.

ETLs for Jassid Management

The economic threshold level (ETL) is defined as “the pest density at which control measures should be applied to prevent an increasing pest population from reaching the economic injury level (EIL)” (Pedigo, Hutchins, & Higley, 1986). The EIL represents the lowest pest density that will cause economic damage, while the ETL serves as a practical decision point for initiating pest management to avert yield loss. Thus, ETLs bridge the gap between economic injury and preventive intervention, ensuring that pest control actions are both biologically justified and economically sound.

In the case of cotton jassid (*Amrasca biguttula*), ETLs have been established in several cotton-growing regions, including India, Pakistan, and parts of Africa. Adults and nymphs feed on the undersides of leaves using their piercing-sucking mouthparts, resulting in “hopper-burn.” This manifests as chlorotic or necrotic spots, which may coalesce into a mosaic or lead to a scorched appearance of the foliage. The characteristic symptoms include upward curling of leaf margins, defoliation, brittle leaves, stunting of plant growth, and premature abscission of buds, flowers, and fruits. Such damage severely compromises plant vigor and yield potential. In India, national and state-level recommendations generally suggest insecticidal control when an average of one to two jassids per leaf are observed, or when characteristic “hopper-burn” symptoms appear on 50% of plants in a field. Pakistan follows similar guidelines, with recommended thresholds often expressed in terms of one to two nymphs per leaf. In other countries, particularly those outside Asia where *A. biguttula* has recently invaded, thresholds remain less well standardized, with management often triggered once visible leaf damage is evident on young plants.

Surveys for damage symptoms, followed by scouting for adults and nymph counts can help in arriving at ETLs. While deriving actionable thresholds based on ‘hopper-burn index’ could be relatively feasible, applying insect-count-based ETLs for jassid management has serious limitations. First, different cotton genotypes vary in their tolerance to jassid feeding, with some varieties expressing greater physiological resilience or morphological resistance (e.g., hairiness) at similar insect densities. Second, field-based insect counts are inherently difficult: adults are highly mobile, escaping at the slightest disturbance, while nymphs are small, cryptic, and usually concealed on the underside of leaves, preferring to cluster on the adaxial side of the leaf near the proximal end, making them difficult to locate and enumerate. Third, pest infestations are rarely evenly distributed across cotton fields, leading to sampling biases and unreliable estimates of true population pressure. In addition, the efficacy of insecticides varies depending on jassid developmental stage, formulation, and local resistance status, while natural enemies such as predators and parasitoids exert variable levels of suppression that complicate density-based thresholds. Cost considerations further affect farmer decisions, as the relative expense of interventions versus expected yield gains can differ substantially across regions and production systems.



Image source: Keshav Kranthi

Figure-20 Several jassid nymphs on underside of the leaf

For these reasons, damage-based thresholds—for example, the proportion of plants showing hopper-burn symptoms—provide a more practical and biologically relevant basis for management decisions than direct insect counts (Kranthi, *et al* 2010).

Damage symptoms are readily visible, more consistently linked to yield loss, and account for varietal tolerance differences and natural control influences. Consequently, hopper-burn-based thresholds are increasingly favored in extension guidelines, as they integrate pest pressure with crop response, leading to more reliable and farmer-friendly recommendations for jassid management.

Table-5. Characteristics of different life stages of *Amrasca biguttula* on okra and cotton

Country/Region	Recommended ETL	Basis of Threshold	Reference
India	1–2 jassids (nymphs or adults) per leaf; or 50% of plants showing hopperburn symptoms	Insect counts and damage symptoms	ICAR-CICR, 2017; AICCIP Annual Reports
Pakistan	1–2 nymphs per leaf (usually based on 3 fully expanded leaves from top canopy)	Insect counts	Ahmad <i>et al.</i> , 1985; PARC Guidelines
Bangladesh	1–2 nymphs per leaf; hopperburn symptoms on 25–30% of plants	Insect counts and visual symptoms	CDB, 2019 (Cotton Development Board)
Sudan / West Africa (recent reports)	No fixed ETL; action typically initiated when hopperburn symptoms are visible on young plants	Damage symptoms (qualitative)	CABI, 2024; Local extension recommendations
General Extension Guidelines	Hopperburn symptoms visible on 50% of plants trigger insecticidal sprays	Damage-based threshold	FAO Plantwise Datasheets; National Extension Publications

Use of Traps for *Amrasca biguttula*

The deployment of traps, particularly sticky traps with varying colors and natural essential oil treatments, offers an effective strategy for monitoring and managing *Amrasca biguttula* populations in crops like okra (*Abelmoschus esculentus*) and cotton (*Gossypium hirsutum*), enhancing integrated pest management (IPM) through targeted pest attraction with minimal impact on natural enemies (Madhu *et al.*, 2022; Patel, 1994; Raja & Arivudainambi, 2004). The efficacy of these traps depends on color contrasts and attractant properties, providing a non-chemical control option.



Image source: Keshav Kranthi

Figure-21 Yellow sticky trap in a cotton field

Field experiments conducted in India evaluated the attractiveness of sticky traps with contrasting color borders to *A. biguttula* (Raja & Arivudainambi, 2004). Traps painted yellow with a red border captured the highest number of leafhoppers (89.82 per trap), while red traps recorded the lowest (18.07 per trap).

Other configurations, including yellow with green border, green with yellow border, green with red border, and red with yellow border, showed intermediate efficacy, suggesting that yellow-based traps with red borders are most effective due to enhanced visual contrast (Raja & Arivudainambi, 2004). In a separate study on okra, Patel (1994) found

that rectangular plastic traps painted Vanderpoel's green, installed at 20–40 cm height, outperformed pinard yellow traps in capturing jassid adults, indicating green's superiority for this crop under specific conditions.

More recent research by Madhu *et al.* (2022) explored the use of yellow sticky traps treated with six natural essential oils (NEOs)—sandalwood, basil, grapefruit, rose, clove, and mint—during 2016–2018 in cotton fields. Traps treated with sandalwood and basil oils attracted significantly higher numbers of *A. biguttula* and *Bemisia tabaci* compared to untreated traps, with an Attractive Index (AI) greater than 1, indicating strong efficacy. GC-MS analysis identified key volatile compounds: santalene, funebrene, and pentadecane in sandalwood; and linalool, β -farnesene, caryophyllene, and methyl eugenol in basil oil, which likely drive pest attraction. Notably, these treated traps captured fewer natural enemies, suggesting a selective effect that could preserve beneficial insects (Madhu *et al.*, 2022).

These findings highlight that yellow sticky traps with red borders or green traps, enhanced with sandalwood or basil oil treatments, are promising tools for *A. biguttula* monitoring and control.

Oviposition Preference and Deterrents

The oviposition behavior of *Amrasca biguttula* (Ishida, 1913) (Hemiptera: Cicadellidae: Typhlocybinae), historically referred to as *A. devastans* in some studies, is influenced by host plant susceptibility, leaf vein morphology, and volatile compounds, offering insights for managing its pestilence on crops such as cotton (*Gossypium hirsutum*), okra (*Abelmoschus esculentus*), and alternative hosts (Agarwal & Krishnananda, 1976; Saxena & Basit, 1982). These factors, including plant volatiles and physical barriers, can deter oviposition or guide egg-laying preferences, supporting integrated pest management (IPM) strategies.

Studies on cotton (var. PS-10) demonstrated that volatiles from certain plants inhibit *A. biguttula* oviposition when presented at a distance from the ovipositional substrate, without affecting adult arrival or stay (Saxena & Basit, 1982).

The inhibitory effectiveness of plant volatiles decreased in the order: eucalyptus > coriander = castor = tomato > lime, with *Ocimum* showing no effect. Among volatile chemicals, inhibitory effects ranked as: citral = carvacrol > citronellol = farnesol = geraniol = eucalyptus oil > neem oil = Cymbopogon oil. Carvacrol exhibited a slight toxic effect on nymphs, but none of the volatiles were toxic to adults, suggesting their role as anti-ovipositional agents rather than insecticides (Saxena & Basit, 1982).

Host plant resistance also shapes oviposition preferences. *A. biguttula* preferentially oviposits on susceptible cotton varieties over medium-resistant or highly resistant ones, with egg densities highest on main leaf veins (Agarwal & Krishnananda, 1976). Leaf vein morphology and trichome characteristics further modulate oviposition. Thicker midribs and longer veins positively correlate with egg-laying, with correlations of $r=0.60$ (main vein), $r=0.65$ (lateral veins), and $r=0.56$ (sub-veins) reported (Arvind Sharma & Ram Singh, 1999; Yadava *et al.*, 1966). In okra, sub-veins received the most eggs, followed by lateral and main veins, attributed to thicker veins across all categories compared to cotton, castor, cowpea, aubergine, and country mallow (Arvind Sharma & Ram Singh, 1999). However, trichome density and length showed no significant correlation with egg numbers, despite earlier reports suggesting that increased hair length and density on cotton midribs deter oviposition (Batra & Gupta, 1970; Balasubramanian *et al.*, 1978; Singh & Agarwal, 1988a). Higher leaf silica content negatively correlated with leafhopper incidence and egg counts on cotton and okra genotypes, enhancing resistance (Singh, 1988; Singh & Agarwal, 1988b).

These findings indicate that *A. biguttula* favors thicker, longer veins on susceptible hosts for oviposition, while plant volatiles (e.g., citral, carvacrol) and physical traits (e.g., silica, trichomes) can deter egg-laying. Deploying resistant varieties with high silica or trichome density, alongside volatile-based deterrents, could reduce oviposition during peak seasons.

Identification of Resistant Genotypes / Varieties

The extensive screening of cotton genotypes over past decades, particularly in India, Pakistan and Myanmar, has successfully identified varieties resistant to *Amrasca biguttula*, with resistance attributed to both genetic and morphological traits. The cultivation of these jassid-resistant varieties is a cornerstone of full-season Integrated Pest Management (IPM). Early in the season, jassids can inflict significant damage on susceptible seedlings through sap-sucking, often necessitating insecticide applications for crop protection. However, these broad-spectrum insecticides, when applied early, are profoundly disruptive; they decimate populations of beneficial predators and parasitoids that are just beginning to establish. Unlike pest populations, which can resurge rapidly, natural enemy communities recover slowly. This early-season disruption cripples the foundation of biolog-

ical control, leading to a loss of its season-long regulatory services and often triggering a dependency on repeated pesticide applications. Therefore, by obviating the need for early-season insecticides, jassid-resistant cultivars preserve and consolidate the naturally occurring biological control from the outset, thereby establishing a stable ecological foundation upon which a successful, season-long IPM program can be built.



Image source: Keshav Kranthi

Figure-22 A healthy cotton field of a jassid resistant variety



Image source: Keshav Kranthi

Figure 23 A field of the resistant variety NDLH 1938

Pushpam and Raveendran (2005) evaluated 13 *Gossypium hirsutum* lines and 30 hybrids, identifying KC 2 as highly resistant, DHY 286, and Khandwa 2 as resistant, and MCU 7 as highly susceptible. These resistant genotypes (KC 2, DHY 286, Khandwa 2) were recommended as donors for breeding jassid-resistant varieties. Kanwat *et al.* (2001) screened 42 elite cotton germplasm entries, with NZC-453, NZC-454, NZC-460, RS-970, and RS-990 (compact and early-maturing types) showing moderate resistance (jassid injury grade of 2).

Imtiaz *et al.* (2002) found genotypes CRIS-129, CRIS-121, CRIS-124, Red okra, CRIS-128, AEH-2, and CRIS-9 exhibited a medium response to jassid attack. Syed *et al.* (2003)

reported the lowest jassid population (2.06/leaf) on variety Rajhans. Chandramani *et al.* (2004) screened 466 cotton breeding entries across multiple seasons, identifying resistant genotypes including Anjali, TKKH 1, TKH 1179, PKV Rajat, TSH 9417, ARB 8824, CNO 6, CNH 1025, CPD 612, MCU 11, SVPR 2, and SVPR 3 (summer 2001); SVPR 3, MCU 11, Anjali, TKKH 1, TKH 1179, TSH 9417, 9804, 8219, 8235, and 8352 (summer 2002); RAH 111, CNH 2124, RHC 940, RAC 9553, PKV, and Rajat (summer 2003); and SVPR 2, TSH 9704, GJHV 392, GJHV 360, GSHV 97/13, RAC 1094, GSHH 19/59, and CNH 301.

Muhammad *et al.* (2009) assessed five cotton cultivars, finding FH-634 most resistant to the sucking pest complex (whitefly, thrips, jassid, aphid), while FH-682 was the most resistant to jassids. Khan *et al.* (2011) evaluated *Bt* cotton genotypes, with IR-443 showing the lowest jassid, thrips, and whitefly infestation, followed by IR-FH-901, indicating excellent overall performance. Salman *et al.* (2011) identified NIAB-86, MNH-63, and SLH-257 as highly susceptible, while NIAB Karishma and CIM-482 showed medium resistance to jassid attack.

Manivannan *et al.* (2021) screened 54 genotypes with resistant check NDLH1938 and susceptible check DCH32, selecting 21 for further studies. Field screening identified nine resistant genotypes (AKH1355, GISV 216, AKH 2012-8, GSHV 173, GISV 267, AKH 1301, GSHV 171, NDLH 2010, AKH 2006-2) on par with NDLH1938, while host preference and nymphal emergence tests confirmed seven (RS 2711, GISV 267, LHDP 1, AKH 1355, RS 2765, F 2164, GISV 216). Mawblei *et al.* (2024) evaluated 100 genotypes with five checks, identifying 19 resistant genotypes comparable to check KC 3.

Sasikumar and Radhika (2020) screened 67 genotypes, classifying 9 as highly resistant (e.g., NDLH 1938, RB 615, RHC 1409), 29 as resistant, 25 as susceptible, and 4 as highly susceptible (0.57–5.83/3 leaves/plant). Madhu *et al.* (2024) found NDLH1938, Suraksha, and F1 hybrids TVH002 × Suraksha, NDLH1938 × Suraksha, and TVH002 × NDLH1938 significantly resistant ($p \leq 0.05$) due to lower leafhopper populations, injury, susceptibility indexes, and host preference survival rates.

Maw *et al.* (2020), Chan Myae Aung *et al.* (2015), and Mi *et al.* (2020) assessed cotton varieties in Myanmar, identifying Line-1, Line-66, Line-96-74-10, RAKA-666, and SD-2014 as moderately resistant due to high leaf trichome density and low Host Borer Index (HBI), while varieties like LGNC-4, LNC-496, and Shwe Daung-8 were susceptible due to low trichome density. Senguttuvan *et al.* (2022) classified TCH 357, TCH 1809, TCH 1895, TCH 1897, TCH 1941, TCH 1828, TSH 383, TVH 002, TVH 003, TKH 0762, SVPR 6, and CO 15 as moderately resistant (2.75–4.42/3 leaves), with KC3 as resistant. Patel and Radadia (2018) designated 11 varieties (e.g., G.Cot.-12, GISV267) as moderately resistant (2.41–6.37/3 leaves).

Additional studies reinforce these findings. Ali *et al.* (1995) noted CIM70 as susceptible (1.69/leaf) and P-43/60 as resistant (0.71/leaf). Murugesan and Kavitha (2010) categorized KC 2 and SVPR 2 as highly resistant, TKH 1128 as resistant, and MCU 5, MCU 10, NISD 2, TKH 1143, TKH 1175 as intermediate. Rohini (2010) identified RAH-100 as highly resistant to leafhoppers and LK-861 as resistant to multiple pests. Amjad *et al.* (2009) found FH-682 and FH-634 resistant. Nawanich *et al.* (2008) reported SRI-1 with the lowest leafhopper density. Vikas *et al.* (2007) noted H-1246 and Ratna as resistant among 346 genotypes. Khan *et al.* (2003) identified Ravi as the most resistant (1.27/leaf), while Syed *et al.* (2003) found Greg-25V most susceptible (2.72/leaf). Gururaj *et al.* (2002) ranked H 1226 as least preferred. Nizamani *et al.* (2002) listed CRIS-129, CRIS-121, CRIS-124, Red okra, CRIS-128, AEH-2, and CRIS-9 as resistant. Malik and Nandal (1986) observed lower jassid incidence in *G. arboreum* (8.7–12.4 nymphs/15 leaves) than *G. hirsutum* (12.4–16.2 nymphs/15 leaves).

The following cotton genotypes have been identified as highly resistant or resistant to *Amrasca biguttula* based on extensive screening studies, making them valuable sources or parents for breeding programs aimed at enhancing jassid resistance.

Highly resistant genotypes include AKH 1355, Anjali, FH-634, IR-443, KC 2, NDLH 1938, RAH-100, and Suraksha, noted for their significantly low leafhopper populations, injury indexes, and host preference survival rates.

Resistant genotypes include AEH-2, AKH 1301, AKH 2006-2, AKH 2012-8, ARB 8824, BGDS 1063, CNH 1025, CNH 2124, CNH 301, CNO 6, CPD 612, CRIS-121, CRIS-124, CRIS-128, CRIS-129, CRIS-9, DHY 286, F 2164, FH-682, GISV 216, GISV 267, GISV 323, GJHV 360, GJHV 392, GJHV 566, GJHV-554, GSHH 19/59, GSHV 171, GSHV 173, GSHV 97/13, GTHV 15-34, H-1246, IR-FH-901, J-129, KC 3, Khandwa 2, LHDP 1, LK-861, LRA 5166, MCU 11, NDLH 2010, P-43/60, PKV 081, PKV Rajat, RAC 1094, RAC 9553, RAH 111, RAHC 1028, RAHH 1951, Rajat, Rajhans, Ratna, Ravi, RB 615, Red okra, RHC 1409, RHC 940, RS 2711, RS 2765, SRI-1, Stoneville, SVPR 2, SVPR 3, SVPR 6, TKH 1128, TKH 1179, TKKH 1, TSH 9417, TSH 9704, TVH002, ZC (BGDS 1063), B 1007, Bar-7-8-2, F 414, Fateh, F 891, Gcot-10, Gcot-12, Jhurar, 2341-0R, J-150, LD 230, LD 237, Pantose 4, RS 992, Reba-B-50, AKH-9015, GISV-69, KH-121, GJHV-217, CA-899, PUSA-4515, GBHV-139, CCH-2584, KH-211, NH-514, RAH-2211, KH-134, ARB-2002, IH-63-MR, AKH-8828, CJHS-52, JLH-1981, KH-107, PH-92-260, JLH-1294, KH-114-4, CICR-HH-1, CCH-510-4, and KR-13. These genotypes exhibit traits such as high trichome density, elevated gossypol gland content, and low susceptibility indexes, offering a robust foundation for developing jassid-resistant cultivars. Further, several wild cotton species are recognized as important sources of resistance to *Amrasca biguttula*, including *G. anomalum*, *G. armourianum*, *G. raimondii*, and *G. tomentosum*.

Hairiness & Gossypol as Resistance Mechanisms

Numerous studies report a significant negative correlation between trichome density and jassid populations. Cotton resistance to *Amrasca biguttula*, is strongly linked to morphological traits such as trichome (hair) density, length, and structure, alongside biochemical factors like gossypol glands, providing a dual defense through antixenosis (detering settling and oviposition) and antibiosis (impeding development and survival). These traits, particularly prominent on leaf surfaces, are critical for breeding resistant cultivars that would lay the foundation for integrated pest management (IPM) strategies (Madhu *et al.*, 2024; Tidke & Sane, 1962).

Correlation analyses show trichome density on midribs and veins negatively impacting jassid numbers per leaf (r significant, negative) (Aheer *et al.*, 1999; Amjad *et al.*, 1999; Ashfaq *et al.*, 2010; Bhatti *et al.*, 2015; Gonde *et al.*, 2015; Gulzar & Sanpal, 2005; Hassan *et al.*, 1999; Khan *et al.*, 2017; Khurshid *et al.*, 2023; Rustamani *et al.*, 2014; Sankeshwar *et al.*, 2016). Bhatti *et al.* (2015) reported trichome length correlat-

ed positively with density ($r=0.875$), abaxial leaf pubescence ($r=0.899$), midrib pubescence ($r=0.678$), vein pubescence ($r=0.814$), and gossypol glands ($r=0.934$), while density correlated with abaxial pubescence ($r=0.835$), vein pubescence ($r=0.742$), and gossypol glands ($r=0.720$). Abaxial pubescence further correlated with midrib pubescence ($r=0.791$), vein pubescence ($r=0.865$), and gossypol glands ($r=0.838$), and vein pubescence with midrib pubescence ($r=0.819$) and gossypol glands ($r=0.773$). Madhu *et al.* (2024) confirmed glandular and non-glandular trichomes via stereo and FE-SEM, with negative correlations between trichome length ($r=-0.52$) and density ($r=-0.72$) with jassid populations.

Species and varietal preferences highlight these traits' efficacy. *Gossypium arboreum* varieties are less preferred than *G. hirsutum*, with resistance in the latter linked to long hair length and tough ventral leaf laminae (Balraj Singh *et al.*, 1976; Khan & Agarwal, 1984; Malik & Nandal, 1986; Premsekhar, 1985; Yein, 1983). Resistant *G. hirsutum* genotypes



Figure-24 Jassid on the abaxial side of glabrous leaves



Figure-25 Jassid on hairy leaves



Figure-26 Jassid on leaves having dense gossypol glands



Figure-27 Jassid on a hairy leaf having dense gossypol glands

like Rupali, CRIS-467, and CRIS-134 exhibit higher trichome density and lower jassid populations (Ali *et al.*, 1995; Ahmad *et al.*, 2004; Sajjad *et al.*, 2004). Rush-pam and Ravikesavan (2019) found resistant parents KC 2, Stoneville, and DHY 286 with higher adaxial trichome and gossypol gland densities, with significant negative correlations ($r=-0.65$ for trichome density, $r=-0.37$ for gossypol gland density) with susceptibility. Varieties with ventral midrib hair exceeding the jassid ovipositor length (e.g., Sanguineum, Sujay, Badnawar-1, M-495) were least preferred for oviposition (Khan *et al.*, 1984). Naveed *et al.* (2011) found a positive correlation between leafhopper population and higher hair density (1011 ± 21.04) with shorter stature (644 ± 27.3), while Ashfaq *et al.* (2010) reported thick leaf lamina as a resistance factor, with no significant effect from midrib and vein hair density.

Trichome distribution and pubescence vary across plant parts. Higher hair density on ventral surfaces and mid-veins reduces oviposition and damage (Manivannan *et al.*, 2017; Murugesan & Kavitha, 2010; Uthamasamy, 1985; Mohankumar, 1996), with upward increases on new leaves enhancing resistance (Madhu *et al.*, 2024). Chan Myae Aung *et al.* (2015) noted genotypes 96-74-10, Line 66, and Lungyaw 3 with high trichome density showed lower jassid incidence. However, excessive pubescence may increase susceptibility to whiteflies and thrips, indicating trade-offs (Barroga *et al.*, 1993; Bhat *et al.*, 1981; Syed *et al.*, 1996).

Gossypol glands complement hairiness. Higher gland density on midribs, veins, and laminae correlates positively with resistance traits ($r=0.720-0.934$) and negatively with oviposition and nymphal emergence (Muhammad *et al.*, 2004; Rushpan & Ravikesavan, 2019). Ramamoorthy *et al.* (2006) found introgression from wild species (*G. raimondii*, *G. armourianum*) introduced heritable hairiness and citronella-like odors, enhancing resistance. Khan *et al.* (2017) reported linear regression models indicating hair density on laminae and gossypol glands on midribs account for 98.2% of jassid population impact.

Breeding implications are significant. High trichome density (e.g., $56/300 \mu\text{m}^2$ in KC3) and moderate hairiness reduce jassid pressure, as seen in resistant hybrids like Ajeet-155 ($51/0.45 \text{ cm}^2$ leaf disc) versus susceptible Siddu ($30/0.45 \text{ cm}^2$) (Manendra, 2012; Senguttuvan *et al.*, 2022). Optimal hair density balances jassid resistance with yield, avoiding adverse agronomic effects (Muhammad *et al.*, 2011). Bt cotton lines can integrate these traits to mitigate jassid increases (Ashfaq *et al.*, 2010; Irfan *et al.*, 2010) thus providing comprehensive resistance to jassids and bollworms. However, nuanced interactions exist, where trichome density may not reduce hopper abundance but lowers injury (Raza *et al.*, 2000; Kaplan *et al.*, 2009).

Leaf Anatomical Resistance

Studies showed that nymphal survival varied significantly: 92–96% on susceptible varieties, 75% on medium-resistant, and 33–50% on resistant varieties, with nymphal periods extended by 3–4 days on resistant types due to antibiosis mechanisms (Agarwal & Krishnananda, 1976). No size differences were noted within the same instar across varieties, but inter-instar size varied, reflecting host influence on development (Agarwal & Krishnananda, 1976). On other hosts, survival was highest on cotton (83.33%) compared to alternative host crops (Sharma, 1996).

Anatomical traits of host plants, particularly in cotton (*Gossypium hirsutum*) and okra (*Abelmoschus esculentus*), confer resistance to *Amrasca biguttula* (Ishida, 1913) (Hemiptera: Cicadellidae: Typhlocybinae) by impeding oviposition, feeding, and nymphal development, providing a foundation for breeding resistant cultivars within integrated pest management (IPM) frameworks (Esquivel *et al.*, 2025). These traits, including leaf thickness, trichome density, vein structure, and glandular compounds, interact complexly to influence pest preference and survival (Kulkarni & Khadi, 2009; Manivannan *et al.*, 2021).

Leaf thickness and compactness are key resistance factors. Thicker leaf laminae, as observed in 30 cotton accessions, correlated positively with jassid resistance, with compact arrangements of lower epidermal, upper epidermal, and mesophyll cells reducing pest population buildup (Tidke & Sane, 1962). In resistant genotypes, midrib cortical cells are densely packed with minimal intercellular space, lower-

ing the piercing rate and phloem injection by *A. biguttula*, thus enhancing resistance (Kadapa *et al.*, 1988). Conversely, succulent thick leaves, while facilitating feeding due to easier stylet penetration, are preferred by the pest, indicating a trade-off (Manivannan *et al.*, 2021). Studies on inter- and intra-specific cotton hybrids showed a high positive correlation (r significant) between nymphal population and cortex thickness, with thinner cortex and vein diameters associated with resistance (Yadava *et al.*, 1967). Leaf area also plays a role, as larger surfaces increase susceptibility by providing more oviposition sites (Batra & Gupta, 1970). Further, long internodes in variety HB 69 contribute to resistance (Balasubramanian, 1979).

Trichomes and glandular structures further deter *A. biguttula*. Non-glandular trichomes interfere with insect movement and access to the leaf epidermis, complicating feeding (Southwood, 1986). Higher trichome density and hair length on midribs reduce oviposition, though excessive pubescence may negatively affect agronomic performance (Balasubramanian *et al.*, 1978; Kulkarni & Khadi, 2009; Singh & Agarwal, 1988a). Genotypes with elevated trichome and gossypol gland densities exhibit reduced jassid emergence, shorter nymphal periods, and lower survival rates (Javed *et al.*, 1992; Muhammad *et al.*, 2004). However, non-pubescent varieties are not inherently susceptible if coupled with thick laminae and dense cortical cells, which act as physical barriers (Kulkarni & Khadi, 2009). Higher silica content in leaves also negatively correlates with leafhopper incidence and egg numbers, reinforcing resistance (Singh, 1988; Singh & Agarwal, 1988b).

Vein morphology and anatomical distances influence pest behavior. Thicker midribs and longer veins positively correlate with oviposition ($r=0.60$ for main veins, $r=0.65$ for lateral veins), with okra's thicker veins across all categories (main, lateral, sub-veins) stimulating higher egg-laying compared to cotton or castor (Arvind Sharma & Ram Singh, 1999). The sucking distance (0.465 mm from lower midrib epidermis to phloem) is shorter than the jassid's stylet length (0.526 mm), but resistance hinges on dense cortex cell arrangement rather than distance alone (Kulkarni & Khadi, 2009). Parameters like phloem bundle thickness and distance from the lower epidermis to phloem pose obstacles to stylet penetration, affecting feeding success (Manivannan *et al.*, 2021).

Additional traits, such as okra leaf type and wild species introgression, enhance resistance. Okra leaf isogenic lines show reduced jassid numbers and higher yields compared to regular-leaf types due to altered morphology (Ali *et al.*, 1993). Breeding efforts have incorporated trichome-based resistance and glandular odors from wild diploids (*G. raimondii*, *G. armourianum*) into *G. hirsutum*, significantly boosting resistance mechanisms (Ramamoorthy *et al.*, 2006). Morphological traits like leaf texture and glandular structures (e.g., gossypol glands) further contribute, with variability across cultivars reflecting complex trait interactions (Sivasubramanian *et al.*, 1991).

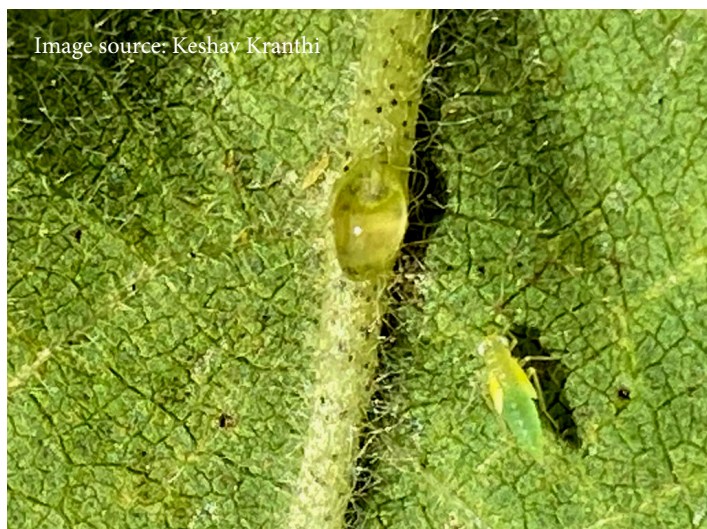


Image source: Keshav Kranthi

Figure-28 Nectary on the abaxial side of a cotton leaf

Biochemical Resistance

Biochemical traits in cotton and related species confer resistance to *Amrasca biguttula* by deterring oviposition, feeding, and development, offering a foundation for resistant cultivar development within integrated pest management (IPM) programs. Compounds such as tannins, gossypol, phenols, and silica, alongside nutrient profiles, influence pest susceptibility, with resistant genotypes exhibiting distinct chemical compositions (Madhu *et al.*, 2024; Singh *et al.*, 1971).

Singh *et al.* (1971) identified 11 resistant upland cotton strains, noting higher tannin and silica contents reduced *A. biguttula* feeding, while susceptible varieties were rich in reducing sugars, calcium, protein, lime, and sulfate. Jassids preferred lipids and proteins, suggesting these nutrients enhance host suitability. Singh and Agarwal (1988a) confirmed reducing sugars, tannins, silica, and free gossypol in cotton leaves showed significant negative correlations with egg numbers, reinforcing their role as feeding deterrents. Peroxidase activity and soluble protein levels serve as resistance indices, with higher peroxidase activity indicating reduced jassid susceptibility (Bhat, 1981; Bhat *et al.*, 1981).

Phenolic compounds and allelochemicals in cotton were found to modulate resistance to jassids. Rohini *et al.* (2011) found genotypes RAH-100, 4085, DHY-286 Sel, and G.cot 16 exhibited low injury indices due to elevated tannins, phenols, and gossypol, while LK-861, Mallika non-Bt, Acala-2, CNHPY-6, and CPD-431 showed resistance to both jassids and whiteflies. Senguttuvan *et al.* (2022) reported resistant genotype KC3 with the highest phenol (4.3 mg g⁻¹), amino acids (136 mg g⁻¹), and tannin (167 mg g⁻¹), contrasting with susceptible DCH 32's lower levels (1.2 mg g⁻¹ phenol, 18 mg g⁻¹ amino acids, 40 mg g⁻¹ tannin). Mandhania *et al.* (2016) noted total phenol, condensed tannin, and total gossypol contents showed significant negative relationships with jassid incidence across growth stages (65–95 DAS), with PUSA 5760 and CA 105 displaying the highest allelochemicals and lowest sugars. However, Vanitha *et al.* (2007) found no sig-

nificant influence of phenols and tannins on jassid growth, suggesting context-dependent effects.

Nutrient imbalances contribute to susceptibility. Mansoor and Waqas (2000) reported low nitrogen, potassium, zinc, and copper levels increased jassid vulnerability, while high chlorophyll, nitrogen, protein, and amino acids favored infestation (Manivannan *et al.*, 2021). Mawblei *et al.* (2024) found total soluble sugar, protein, and free amino acids positively correlated with jassid infestation grades, whereas phenols and trichome density were negatively correlated. Madhu *et al.* (2024) noted reducing sugars, chlorophyll, and amino acids were positively associated with pest density, while higher gossypol and tannin levels reduced infestations, acting as feeding deterrents. Kanher *et al.* (2018) identified minimum condensed tannin and total gossypol concentrations as key resistance factors, with Rushpam and Ravikesavan (2019) reporting significant negative correlations ($r=-0.32$ for phenol, $r=-0.40$ for gossypol) with susceptibility.

Historical studies align with these findings. Sharma and Agarwal (1983) observed negative correlations between jassid populations and leaf thickness, sugars, and tannins, alongside positive associations with protein and moisture (Nizamani *et al.*, 2002). Resistant cultivars BJR and M-495 showed lower pest populations, linked to higher gossypol, sugars, and free amino acids, and lower phenols and tannins, though feeding-induced tannin accumulation suggested secondary resistance factors. Mohan *et al.* (1987) noted phenolics in high concentrations exert direct toxicity, while lipids and minerals in resistant lines bolster defense (Singh *et al.*, 1971). Genotypes with high trichome density and gossypol glands further enhance resistance (Sajjad *et al.*, 2004). The studies indicate that breeding cultivars with high tannin, gossypol, and phenol levels, and low sugars and amino acids, combined with anatomical features that are correlated with resistance coupled with partial hairiness could circumvent the problems associated with highly hairy cotton genotypes and could help in mitigating jassid pest pressure during the season.

Seed Treatment

Seed treatment with various insecticides and biological agents has proven effective in managing *Amrasca biguttula* populations in cotton, while also influencing plant growth and beneficial arthropod dynamics. Saeed *et al.* (2016) demonstrated that neonicotinoid seed treatments, specifically imidacloprid and thiamethoxam, efficiently suppress leafhopper populations under field conditions, with higher dosages yielding greater reductions. Imidacloprid at the recommended dose of 5 g kg⁻¹ seed delayed the pest reaching the economic damage threshold by approximately 20 days, compared to 10–15 days for thiamethoxam, while also enhancing plant growth directly and via pest suppression. However, both treatments reduced beneficial arthropod abundance, with imidacloprid showing no significant negative impact on natural enemies at recommended doses, unlike thiamethoxam, though sublethal effects on individuals were not assessed.

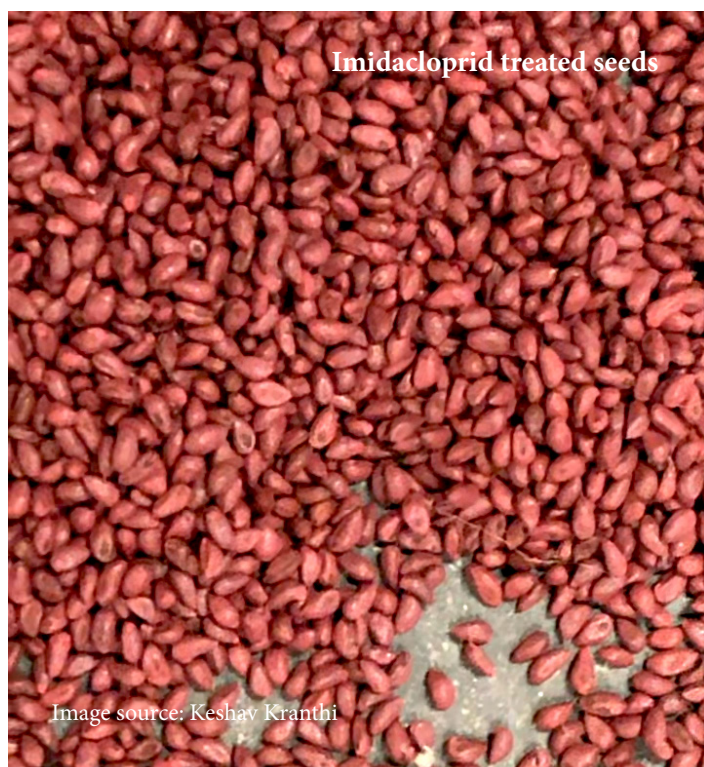


Figure-29 Imidacloprid treated cotton seeds

Satyan *et al.* (2015) evaluated 14 synthetic insecticides for seed treatment, finding acephate to be the most effective, reducing leafhopper populations to 0.70/3 leaves, followed by triazophos (1.70/3 leaves), acetamiprid (1.80/3 leaves), monocrotophos (2.30/3 leaves), imidacloprid 48 FS (2.40/3 leaves), and carbosulfan (2.60/3 leaves), compared to 5.40/3 leaves in untreated plots by the fourth week after sowing. Imidacloprid 48 FS also maximized germination (48.81%) and improved shoot length (10.60 cm), while carbosulfan enhanced root length (20.67 cm).

Murugesan and Kavitha (2009) tested imidacloprid, monocrotophos, *Pseudomonas fluorescens*, and other treatments, with imidacloprid proving most effective, reducing leafhopper populations to 0.8/3 leaves (research farm) and 0.53/3 leaves (on-farm trial), achieving 72.54% and 59.59% reduction, respectively.

Monocrotophos (1.23/3 leaves) and *P. fluorescens* (1.42/3 leaves) also exceeded 50% reduction, while other treatments like acephate and neem oil were less effective.

Laboratory studies confirmed imidacloprid, monocrotophos, and *P. fluorescens* improved germination and shoot length, though neem oil negatively affected shoot growth.

These findings suggest imidacloprid at recommended doses is a promising seed treatment for managing *A. biguttula*, balancing pest control with plant growth enhancement and minimal disruption to beneficial arthropods, while seed treatment with *P. fluorescens* can be considered as a biological option especially for organic cotton production systems.

Chemical Control

Chemical control remains the predominant strategy employed by farmers to protect cotton crops from *Amrasca devastans* infestations, though its extensive use has led to resistance development, particularly against pyrethroid insecticides (Razaq *et al.*, 2013; Ahmad *et al.*, 1999). Lakshmanan *et al.* (2021) conducted a field experiment at Nandyal, India, during kharif 2017–2018, evaluating insecticides under rain-fed conditions. Monocrotophos (36% SL @ 1.6 ml/L), flonicamid (50% WDG @ 0.3 g/L), and sulfoxaflor (75% WG @ 1.5 ml/L) were most effective against leafhoppers, with monocrotophos yielding the highest seed cotton yield (1376 kg/ha), closely followed by flonicamid (1248 kg/ha) and sulfoxaflor (1235 kg/ha). Ghosh (2020) tested dinotefuran (20 SG @ 40 g a.i./ha), fipronil (5% SC @ 37.5 g a.i./ha), and imidacloprid (70 WG @ 24.5 g a.i./ha), achieving the highest jassid suppression (90.29%, 89.34%, and 78.42%, respectively) and thrips control, while remaining safe for coccinellid predators. Bebagar *et al.* (2023) found imidacloprid (17.8% SL) most effective, reducing jassid populations by 81.25% with a yield of 3200 kg/ha and a cost-benefit ratio (CBR) of 12.57, outperforming thiamethoxam (77% reduction, 3150 kg/ha, CBR 13.38) and lambda-cyhalothrin (72.25% reduction, 3050 kg/ha, CBR 13.12). Diabate *et al.* (2024) reported superior efficacy with insecticides like flonicamid, yielding the highest cotton yields over two seasons (2021–2023), underscoring chemicals' edge in yield optimization.



Figure-30 Pesticide display in store in India

Dake and Bhamare (2019) assessed newer insecticides on sunflower, with imidacloprid (17.8 SL @ 0.003%) showing the greatest efficacy (4.20 and 3.40 jassids/leaf after first and second sprays) and highest persistent toxicity (PT 804.3 and 843.08, LT50 5.75 and 5.91 days), followed by spinosad and indoxacarb. Kumar *et al.* (2019) evaluated insecticides on brinjal, with imidacloprid (17.8 SL @ 100 ml/ha) and fipronil (5 SC @ 750 ml/ha) leading in jassid control. Rekha *et al.* (2017) identified thiamethoxam (25 WDG @ 0.2 g/L, LC50 4.03 ppm) and flonicamid (50 WG @ 0.3 g/L, LC50 4.50 ppm) as top performers against okra leafhoppers in laboratory bioassays, with field trials confirming thiamethoxam (25 g a.i./ha) as most effective, followed by flonicamid (75 g a.i./ha). Hafeez *et al.* (2020) reported nitenpyram as the most effective.

tive (0.23–0.53 jassids/leaf), followed by chlorfenpyr, against cotton jassids, with acephate being ineffective. Saleem *et al.* (2018) found clothianidin (84% reduction), nitenpyram (71%), and momentum (67%) superior to buprofezin (65%) for jassid control, with effectiveness declining over time.

Susheelkumar *et al.* (2020) tested insecticides on okra, with acetamiprid (20% SP @ 0.3 g/L, 83.06% reduction) and imidacloprid (17.8% SL @ 0.5 ml/L, 82.91%) being the most effective, followed by acephate (77.48%). Akbar *et al.* (2012) evaluated biopesticides and conventional insecticides on potato, with imidacloprid (88.59% reduction) outperforming endosulfan (73.32%) and profenofos (64.87%). Sahito *et al.* (2017) found nitenpyram (68.61% reduction) most effective against cotton jassids, followed by acephate (58.75%).

Sharma and Summarwar (2017) reported acephate (75% SP) achieving 76.10% jassid reduction on *Bt* cotton, superior to other treatments. Sarma *et al.* (2016) identified fipronil (5% SC @ 50 g a.i./ha), flonicamid (@ 0.3 g/L), and diafenthiuron (@ 375 g a.i./ha) as effective, with flonicamid yielding 13.33 q/ha. Afzal *et al.* (2014) noted imidacloprid, diafenthiuron, acetamiprid, and thiamethoxam as highly effective, with imidacloprid achieving 92.42% mortality after the first spray. Bharati *et al.* (2015) found imidacloprid (0.004%) and dimethoate (0.03%) most effective against brinjal jassids. Sameer *et al.* (2023) reported sulfoxaflor (3.7%) + bifenthrin (11.2% SC @ 37+114 g a.i./ha) as the best, followed by lower doses of the combination, with cypermethrin least effective. The studies showed that Imidacloprid, flonicamid, sulfoxaflor, and fipronil emerge as consistently effective against *A. devastans*, balancing pest control with yield considerations.

Insecticide Resistance

Studies conducted over the past three decades showed that *Amrasca devastans* has developed significant resistance to various insecticides across India, Pakistan, and other Asian regions due to their widespread and often indiscriminate use in cotton and other crops.

In India, resistance to metasystox, dimethoate, and phosphamidon has been documented (Santhini and Uthamasamy, 1997; Chalam and Subbaratnam, 1999; Chalam *et al.*, 2001; Praveen, 2003), with Jeya Pradeepa (2000) and Jhansi *et al.* (2004) reporting resistance to endosulfan, monocrotophos, cypermethrin, phosphamidon, dimethoate, methyl demeton, and acephate, particularly linked to excessive insecticide application in Andhra Pradesh.

Ram Singh and Jaglan (2005) further noted resistance to fenvalerate, malathion, oxydemeton-methyl, phosphamidon, cypermethrin, deltamethrin, and zeta-cypermethrin. In Pakistan, Mushtaq Ahmad *et al.* (1999) identified high resistance levels to pyrethroids, with zeta-cypermethrin showing a 108.9-fold resistance at LC50, followed by deltamethrin (105.5-fold) and cypermethrin (25-fold).

More recent studies highlight evolving resistance to neonicotinoids and other classes. Saeed *et al.* (2017) monitored six populations in Pakistan, reporting resistance ratios (RRs) of 2.3–29.3-fold for acetamiprid, 4.8–95.0-fold for imidacloprid, and 19.1–1197.9-fold for thiamethoxam compared to a reference population, urging integrated resistance management (IRM) with insecticide rotation and biocontrol.

Hussain *et al.* (2025) assessed nine districts in Pakistan, finding resistance ranges of 10.31–41.23-fold to profenofos, 10.07–83.57-fold to chlorpyrifos, 8.70–30.02-fold to bifenthrin, 4.56–26.24-fold to deltamethrin, 56.28–218.16-fold to imidacloprid, 5.70–23.30-fold to fipronil, 2.99–18.26-fold to clothianidin, 4.66–25.43-fold to chlorfenapyr, and 5.33–44.51-fold to carbosulfan. Mohan *et al.* (2025) noted neonicotinoid resistance in Guntur (Andhra Pradesh), Maharashtra, and Karnataka, with imidacloprid showing the lowest LC50 (24.91 ppm) yet indicating sensitivity limits (Mahalakshmi and Prasad, 2020; Pate *et al.*, 2020; Halappa and Patil, 2016; Arunkumara *et al.*, 2020).

Abbas *et al.* (2018) evaluated four populations in Pakistan, revealing 32.95–136.47-fold resistance to nitenpyram, 23.03–56.74-fold to spirotetramat, 10.84–31.33-fold to chlorfenapyr, 1.20–9.43-fold to fipronil, 3.27–43.77-fold to emamectin benzoate, 0.66–2.81-fold to sulfoxaflor, and 0.59–1.25-fold to flonicamid, recommending discontinuation of resistant insecticides and rotation with effective ones.

Another study by Abbas *et al.* (2018) found 11.10–92.87-fold resistance to deltamethrin, 5.87–14.11-fold to bifenthrin, 3.16–17.5-fold to cypermethrin, 2.65–36.42-fold to chlorpyrifos, 7.28–57.71-fold to profenofos, 1.65–11.13-fold to acephate, and 2.55–43.31-fold to methomyl across six districts, attributing this to injudicious use and advocating for new chemistry insecticides. Pradeepa and Regupathy (2002) in Tamil Nadu reported resistance evolution over generations, with imidacloprid remaining the most toxic (LC50 0.000457 ppm in F1), while dimethoate, demeton-methyl, and acephate showed reduced susceptibility (susceptibility index 0.63–1.10).

This widespread resistance underscores the need for IRM strategies, including rotating insecticides with diverse modes of action, optimizing application rates, and integrating biocontrol. When managing *Amrasca biguttula* through chemical means, it is critical to conserve naturally occurring beneficial insects; therefore, insecticide choices should be selective to minimize harm to these key biological control agents. Chemical insecticides, such as imidacloprid, flonicamid, sulfoxaflor, and fipronil, have demonstrated high efficacy in reducing jassid populations and boosting yields, with imidacloprid (17.8% SL) achieving up to 81.25% reduction and yields of 3200 kg/ha, though resistance to pyrethroids, organophosphates, and neonicotinoids (e.g., 56.28–218.16-fold to imidacloprid) underscores the need for judicious use of insecticides and implementation of insecticide resistance management strategies.

Botanical Pesticides

Botanical pesticides, derived from plant sources, offer an eco-friendly alternative for managing *Amrasca biguttula* infestations in cotton, okra, and other crops, with varying efficacy and economic benefits. Singh *et al.* (2003) conducted an experiment in Faizabad, Uttar Pradesh, India (1999–2000), testing neem-based products (Achook @ 0.7%, Neemarin @ 0.7%, neem seed kernel extract [NSKE] @ 1% and 3%) comparing efficacy with endosulfan (0.07%) on okra. Endosulfan and NSKE (3%) were most effective, with Achook yielding

the highest yield (5006 Kg/ha), though NSKE (3%) offered the best cost:benefit ratio (1:10.70). Patel (1994) observed that neem-based formulations (neemark 1%, repel 1%, NSKE 5%) and *Ailanthus excelsa* (ardusa) leaf suspension (5%) on brinjal and okra reduced jassid nymph emergence and adult populations due to oviposition deterrence, repellence, and growth inhibition, matching the efficacy of monocrotophos (0.04%) and other synthetics when combined with endosulfan (0.035%).



Figure-31 Nectary on the abaxial side of a cotton leaf

Abdullah *et al.* (2017) evaluated neem seed extract (NSE) on *Bt* and non-*Bt* cotton, achieving maximum reductions of 64.94% (*Bt*) and 69.05% (non-*Bt*) for *A. biguttula* at 6% NSE concentration after 168 hours, compared to 44.50% (*Bt*) and 48.48% (non-*Bt*) at 2%. Patel *et al.* (2025) found azadirachtin (0.006%) most effective on okra (1.81 jassids/leaf, ~50% control), followed by tobacco decoction (2%) and NSKE (5%) (2.26 and 2.49 jassids/leaf).

Naik *et al.* (2012) reported NSKE (5%) reducing leafhopper populations to 3.53 and 4.00/3 leaves after first and second sprays, respectively. Solangi *et al.* (2013) noted neem powder (65.46%) and tobacco leaves (63.16%) as effective, while Mahmood *et al.* (2014) found tobacco extract (99.57% reduction) and neem powder (98.18%) superior against jassids. Iqbal *et al.* (2015) observed neem leaf extract (5%) and garlic leaf extract (5%) reducing populations to 6.31 and 6.86/plant, respectively.

Deepika *et al.* (2018) recorded NSKE (5%) as the best (2.77 and 2.31/3 leaves), followed by garlic extract (5%) (3.50 and 2.68/3 leaves). Devra and Kumar (2022) reported azadirachtin (5000 ppm @ 1.5 ml/L, 44.56%) and neem oil (10 ml/L, 40.34%) as effective. Ahmad (2020) achieved up to 70% jassid control with *Azadirachta indica*, *Nicotiana tabacum*, and other extracts, suggesting their IPM potential.

Nadeem *et al.* (2015) found neem oil (4% and 5%) and lambda-cyhalothrin (330 mL/acre) equally effective against jassids, with significant reductions after 48–72 hours, though efficacy declined after 360 hours, with no impact on plant height but improved yield. Srinivasan and Sundara Babu (2000) reported neem products reducing brinjal leafhopper nymphs to 1.44–3.44/plant versus 8.55 in controls after three sprays. Patel and Patel (1996) confirmed neem-based (neemark 1%, repel 1%, NSKE 5%) and *A. excelsa* (5%) suspensions deterred oviposition, repelled jassids, and inhibited growth on okra.

Ghosh and Chakraborty (2015) found *Polygonum hydropiper* (5%) and *Pongamia pinnata* (5%) extracts, alongside spinosad, achieving over 50% jassid mortality, with *Polygonum* exceeding 60% at 3–7 days after treatment (DAT). Harsha and Vijay (2022) noted garlic extract (30 ml/L, 54%) and neem oil (10 ml/L, 49.17%) as top performers on *Bt* cotton, with Ecotin (azadirachtin 5000 ppm @ 1.5 ml/L) maintaining efficacy (44.56%) and boosting yield.

Bebagar *et al.* (2023) reported neem oil (5%) reducing jassid populations by 65.75% with a yield of 2800 kg/ha (CBR 7.39), outperforming garlic extract (10%, 55.25%) and eucalyptus oil (5%, 50%). Bhandari *et al.* (2022) found neem extract (2 ml/L) comparable to chlorpyrifos + cypermethrin (2 ml/L) after multiple sprays, with cannabis extract being the

least effective but still better than controls. A study on drifted brown seaweeds (DBSW) highlighted *Sargassum wightii* (20.62% tannin) and *Stoechospermum polypodioides* (LC50 0.044%) as effective against *A. devastans* via oral and contact toxicity, reducing enzyme activity and body protein, suggesting a sustainable management option.

Microbial Pesticides

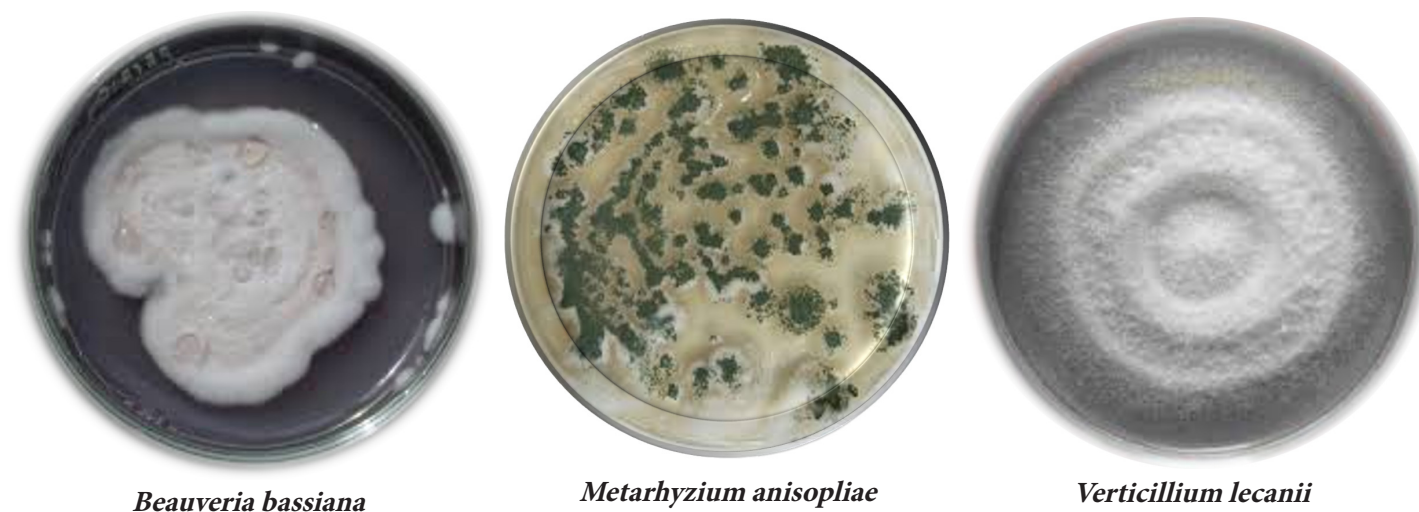


Figure 32 Examples of entomopathogenic fungal species for jassid management

Microbial pesticides, leveraging entomopathogenic fungi, bacteria, and biorationals, offer a promising, eco-friendly approach to manage *Amrasca biguttula* on crops such as okra and cotton, with varying efficacy levels.

Bhushan *et al.* (2023) found *Beauveria bassiana* (1×10^8 conidia/ml @ 10 ml/L) and NSKE (1% @ 2 ml/L) most effective against jassids, with NSKE and spinosad (45% SC @ 0.4 ml/L) supporting predatory coccinellids. Janghel (2015) observed acetamiprid reducing jassid incidence from 4.53 to 1.28/plant (90.59%), with *M. anisopliae*, *C. zastrowi sillemi*, *B. bassiana*, and *V. lecanii* achieving 74.85–58.84% reduction on okra.

Hamad *et al.* (2022) reported a commercial *B. bassiana* isolate achieving 75.7% mortality after 10 days (LT50 6.01 days), outperforming a local isolate (66.7%, LT50 6.94 days) and *Isaria fumosorosea* (62.69%, LT50 7.14 days), with nutrient-enhanced suspensions reducing LT50 further. Nguyen *et al.* (2023) demonstrated *Purpureocillium lilacinum* PL1 (1×10^7 conidia/ml) reducing nymph populations by 88.66% in vitro and 72.87% in okra fields after two applications.

Manjula *et al.* (2018) found soil and foliar *Pseudomonas fluorescens* (1%) achieving 71.00–91.58% reduction on *Bt* and non-*Bt* cotton, boosting yields. Hussein Ali Mutney *et al.* (2020) noted spinosad (63.90% nymph mortality) and *Lecanicillium muscarium* (51.9%) excelling on okra. Lal and Dhurve (2024) reported NSKE (5%) as most effective, followed by neem oil (5%) and *V. lecanii*, with a yield of 119.56

q/ha. Botanical pesticides, including neem-based products (e.g., NSKE at 5% with 65.75% reduction) and seaweed extracts (*Sargassum wightii* with LC50 0.007%), offer sustainable alternatives with moderate efficacy (up to 70% control), lower environmental impact, and support for IPM, though yields with botanical pesticides generally lag behind chemicals.

Manivannan *et al.* (2018) identified *L. lecanii* (1×10^8 spores/ml) with 85.00% mortality (LT50 115.37 hours) and 69.17% reduction in pot studies. Sivakumar *et al.* (2017) discovered *Bacillus pumilus* in jassid guts inhibiting fungal pathogens (e.g., 2.0 cm against *B. bassiana*) and producing digestive enzymes. Maketon *et al.* (2008) found *Metarhizium anisopliae* CKM-048 (1.25×10^{13} conidia/ha) matching lambda-cyhalothrin's efficacy on aubergine.

Sowmiya and Pazhanisamy (2019) found ginger oil (3%) and NSKE (5%) most effective against bhendi leafhoppers, with *Verticillium lecanii* (0.5%) also showing notable reduction during kharif and rabi 2018. Kumar *et al.* (2021) reported neem oil (3% @ 30 ml/L) achieving 65.76% and 82.84% reduction after first and second sprays, respectively, outperforming *Lecanicillium lecanii*, *Beauveria bassiana*, *Metarhizium anisopliae*, and *Bacillus thuringiensis* on okra.

Elango (2020) identified dinotefuran (20 SG @ 0.30 g/L, 91.34%) and buprofezin (25 SC @ 2 ml/L, 88.36%) as top performers, with ginger + garlic + green chili extract (5%) achieving 77.91% reduction, followed by NSKE (5%) and *L. lecanii* (10 ml/L).

Kekan *et al.* (2022) noted azadirachtin (1 EC @ 0.003%) and *L. lecanii* (5 g/L) reducing populations to 3.07 and 3.48/3 leaves, respectively. Microbial pesticides, particularly *L. lecanii*, *B. bassiana*, and NSKE, provide effective, environmentally safe options for jassid control, supporting IPM.

Mahalakshmi *et al.* (2018) noted flonicamid (50% WG @ 75–100 g a.i./ha), diafenthiuron (50% WP @ 300 g a.i./ha), and buprofezin (25% SC @ 250 g a.i./ha) achieving over 70% reduction in *Bt* cotton leafhoppers, significantly outperforming NSKE (5%) and entomopathogens like *Verticillium lecanii* and *Metarhizium anisopliae* (below 50% reduction). Patel *et al.* (2017) found imidacloprid (17.8% SL) most effective against okra jassids (1.04/plant) with the highest benefit:cost ratio (1:9.54), followed by acetamiprid (1.69/plant, 1:7.70) and azadirachtin (2.84/plant, 1:5.81), with biopesticides (*V. lecanii*, *M. anisopliae*, *B. bassiana*) lagging behind.

Soaps, Biopesticides vs. Insecticides

The comparative efficacy of soaps, biopesticides, and insecticides in managing *Amrasca biguttula* (Ishida, 1913) (Hemiptera: Cicadellidae: Typhlocybinae) highlights a spectrum of control options with distinct advantages. Selvapriya and Regupathy (2025) demonstrated that potassium salts of fatty acids (49% SL @ 15–18.75 mL/L) achieved 100% mortality of jassids within 3 days after treatment (DAT), matching the performance of azadirachtin (0.5 mL/L) and imidacloprid (17.8% SL @ 0.25 mL/L), positioning soap-based solutions as a potent, non-toxic alternative. Mollah (2024) found sodium lauryl ether sulfate (Fizimite 20%) effective against brinjal leafhoppers, reducing populations by 85.80% and leaf infestation by 77.84%, closely rivaling the chemical insecticide Voliam Flexi 300 SC, suggesting soaps as a viable eco-friendly option. While insecticides provide rapid, high-efficacy control and yield boosts, soaps and biopesticides offer sustainable, lower-toxicity alternatives, though with reduced potency.

Management Strategies

Effective management of *Amrasca biguttula*, relies on an integrated pest management (IPM) framework that prioritizes ecofriendly and environmentally compatible methods to minimize chemical inputs, preserve natural enemies, and reduce resistance risks. Though, the general economic threshold level (ETL) for intervention is 50 nymphs or adults per 50 leaves or 2–3 nymphs per leaf with 25% infested leaves, guiding actions to avoid unnecessary treatments, in developing countries during the early stages of the crop, based on 40 plants sampled per acre, a biological or botanical control intervention is recommended at a hopper burn index value of approximately 1.5. If the index escalates to a value between 1.5 and 2.0, the application of chemical control measures is advised.

Preventive cultural strategies are foundational, including selection of jassid-resistant, which deter oviposition and feeding. Early, synchronized sowing (e.g., April–May in India or adjusted for local dynamics) avoids peak infestation periods (June–August), while avoiding excessive nitrogenous fertilizers prevents succulent growth that attracts jassids, and en-

suring soils are not potassium-deficient enhances plant resilience against hopper-burn. Removal of weeds, crop residues, and alternate hosts from fields, borders, and irrigation ditches eliminates overwintering sites and carry-over sources, disrupting the pest's lifecycle. Crop rotation with non-Malvaceae plants, avoiding sequential planting of hosts like okra (*Abelmoschus esculentus*), eggplant (*Solanum melongena*), or castor (*Ricinus communis*), and intercropping with trap or repellent crops further dilutes pest pressure and promotes biodiversity.

Biological control emphasizes natural enemies and microbial agents for sustainable suppression. Seed treatment with *Pseudomonas fluorescens* and *Beauveria bassiana* (10 g/kg seed) provides early protection, while soil application of neem cake (250 kg/ha) deters nymphs. Releasing *Chrysoperla carnea* (2 grubs/plant weekly) targets nymphs and adults, and conserving predators like lady beetles, lacewings, and spiders through habitat provision reduces pesticide use. Entomopathogenic fungi such as *B. bassiana*, *Metarhizium anisopliae*, *Verticillium lecanii*, and *Purpureocillium lilacinum* achieve 62.69–88.66% mortality in vitro and 72.87% in fields, with nutrient-enhanced formulations shortening LT50 (Hamad *et al.*, 2022; Nguyen *et al.*, 2023). Gut bacteria like *Bacillus pumilus* also inhibit pathogens and aid host nutrition (Sivakumar *et al.*, 2017). Physical methods, including yellow sticky traps (10/acre) for monitoring and trapping adults, light traps for nocturnal capture, and reflective mulches to repel jassids, support non-chemical control in low-input systems (Raja & Arivudainambi, 2004).

Initial reliance on botanical or microbial pesticides preserves naturally occurring biological control, allowing beneficial insects to thrive and provide season-long suppression of jassids and other pests (Kranthi *et al.*, 2009; Kranthi and Russell, 2009; Kranthi and Kranthi, 2010; Kranthi *et al.*, 2010). Neem-based formulations (NSKE 5%, azadirachtin 0.03% @ 2 mL/L, neem oil 10000 ppm @ 1%) achieve 58.84–77.91% reduction, with garlic + ginger + green chili extract (5%) at 77.91% (Deepika *et al.*, 2018; Elango, 2020). These are safe for coccinellids and cost-effective (Harsha & Vijay, 2022). Seaweed extracts (*Sargassum wightii*) reduce enzyme activity and body protein in jassids (Petchidurai *et al.*, 2023). When chemical insecticides are necessary (e.g., populations exceed ETL), prioritize selective, low-toxicity options like flonicamid (50% WG @ 60 g/acre), buprofezin (25% SC @ 400 mL/acre), diafenthiuron (50% WP @ 240 g/acre), thiacloprid (21.7% SC @ 40–50 mL/acre), thiamethoxam (25% WG @ 40 g/acre), or seed treatment with imidacloprid (75 WS or 17.8 SL @ 5–10 g/kg seed), which provide early-season protection with minimal disruption to predators. Rotate modes of action to delay resistance, and apply foliar sprays only during high injury.

In the U.S., where *A. biguttula* invaded 101 counties by September 30, 2025, preliminary trials showed that Bidrin (dicrotophos), flonicamid, thiomethoxam, flupyradifurone, afidopyrophen and sulfoxaflor were effective in controlling

jassids. Avoiding highly hazardous pesticides (HHPs) such as Bidrin (dicrotophos) is critical, as it is highly toxic (WHO Class Ib), persistent, and disrupts non-target organisms, pollinators, and natural enemies early in the season when beneficials are establishing, risking secondary outbreaks, resistance, and ecological harm. Thus, Bidrin use is not preferred for jassids, as relatively better alternatives (WHO class II to III) like flonicamid, dinotefuran, flupyradifurone, afidopyrophen, sulfoxaflor, fluxametamide or biorationals provide comparable control of jassids without broad-spectrum toxicity.

Sustainable management of *Amrasca biguttula* hinges on integrating ecofriendly strategies that enhance crop resilience and preserve natural ecosystems. Early sowing, coupled with jassid-resistant varieties and seed treatments with imidacloprid or thiamethoxam, establishes a strong foundation, while avoiding excessive nitrogenous fertilizers and correcting potassium deficiencies bolster plant vigor. Natural enemy conservation, agroecological practices like crop diversity and agroforestry, and optimized soil/water management reduce plant stress, supporting long-term suppression. Recommendations emphasize weed removal to prevent carry-over, avoiding host intercropping, minimal use of selective insecticides, and adjusting sowing to misalign with peak pest density. Precision technology, including satellite imagery for early stress detection and targeted applications, minimizes chemical inputs. Initially relying on botanical or microbial pesticides preserves beneficial insects, fostering season-long control of multiple pests, while chemical use, if needed, should prioritize safe, selective options like flonicamid or buprofezin to protect natural enemies.

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