A Review of the Economic Impact and Sustainable Strategies for Mitigation of the Cassava Mite Complex[1]

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J. Entomol. Sci. 60(4): 000–000 (Month 2024)
DOI: 10.18474/JES24-55

Abstract The roots of cassava (*Manihot esculenta* Crantz) are an excellent source of dietary starch for millions of people and also serve as raw material for several industrial products. Cassava root production, however, is limited by insect, mite and disease pests. Tetranychid mites (Acari; Tetranychidae), until recently considered a minor pest of cassava, have assumed major pest status contributing up to 50 to 80% reduction in root yields, causing severe economic losses. Globally, numerous species of tetranychids have been reported causing damage to cassava. These include *Tetranychus urticae* Koch (syn = *T. cinnabarinus* [Poisd.]), *T. truncatus* Ehara, *T. neocaledonicus* André, *T. kanzawai* Kishida, *Mononychellus tanajoa* (Bondar) (syn = *M. progresivus* Doreste), *M. caribbeanae* McGregor, *Oligonychus biharensis* (Hirst), *O. gossypii* (Zacher), and *Eutetranychus orientalis* (Klein). Various studies have been undertaken and reported to explore methods (e.g., cultural methods, biological control, botanicals, host plant resistance, conventional chemical insecticides) to mitigate mite damage to cassava and manage their populations. This present work is a review of the pertinent scientific literature on the cassava mite complex, its economic impact, and potential management strategies for the pests.

Key Words cassava, mite, yield loss, management, biological control

Cassava, *Manihot esculenta* Crantz (syn. *Manihot utilissima* Pohl) (Malpighiales: Euphorbiaceae), the single domesticated species from the genus *Manihot,* is a perennial shrub propagated vegetatively and cultivated throughout the tropical lowlands primarily for its starchy roots (Cock 2011, Cock and Connor 2021, Nassar and Ortiz 2007). Native to the western Amazonian Rim of South America and subsequently transported to Africa by European sailors during the 16th Century and later to Asia in the 18th Century (Ceballos et al. 2004, Tomlinson et al. 2018).

The tuber crop is of paramount importance owing to its use as the staple food for over 300–500 million people from subtropical and tropical areas of Asia, Africa, and Latin America (Bellotti et al. 1987, El-Sharkawy 2003, Pujol et al. 2002). Originally

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[1] Received 5 June 2024; accepted for publication 7 July 2024.

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grown under subsistence agriculture, the crop later became a major crop under commercial farming, fulfilling multiple roles, including providing fresh and several fermented as well as unfermented industrial starch-based products (Falade and Akingbala 2010, Parmar et al. 2017) including *gari*, *fufu*, *lafun*, *agbelima*, bread, syrups, beverages, paper, textile, plywood, glue, biofuel, bioplastics and livestock feed (Balagopalan 2001, Li et al. 2017, Nassar and Ortiz 2007). Its crucial role in global agriculture is underscored by being the third most important source of calories and the sixth most important crop in terms of global annual production grown within the tropics (Burns et al. 2010). Cassava is a climate resilient crop with the potential to prosper under adverse conditions, making it a food security source against famine. It demonstrates high energy production efficiency and substantial yield potential, thus requiring less labor per unit of energy harvested (Ceballos et al. 2004, De Bruijn and Fresco 1989, El-Sharkawy 2003).

The world's annual cassava production in 2022 was estimated at 330.40 million tonnes, grown on 32.04 million ha, with a yield of 10.31 tonnes per ha (FAO 2022). In India alone, the national annual cassava production for the fiscal year 2022–2023 was estimated at 6.05 million tonnes from 167.53 thousand ha with a yield of 36.14 tonnes per ha (INDIASTAT 2023).

Cassava production is threatened by approximately 200 arthropod and nematode pest species, including mites (Acari: Tetranychidae), thrips (*Corynothrips stenopterus* Williams) (Thysanoptera: Thripidae), whiteflies (*Bemisia tabaci* Gennadius) (Hemiptera: Aleyrodidae), mealybugs (*Phenacoccus manihoti* Matile-Ferrero) (Hemiptera: Pseudococcidae) (Bellotti and van Schoonhoven 1978), scales (*Stictococcus vayssierei* Richard) (Nassar and Ortiz 2007), lace bugs (*Vatiga illudens* [Drake]) (Hemiptera: Tingidae), burrower bugs (*Cyrtomenus bergi* Froeschner) (Hemiptera: Cydnidae), stem borers (*Chilomima* spp.) (Lepidoptera: Crambidae) (Bellotti et al. 2012c), *Lagocheirus* spp. (Coleoptera: Cerambycidae), *Coelosternus* spp. (Coleoptera: Cerambycidae), hornworms (*Erinnyis ello* L.) (Lepidoptera: Sphingidae), and nematodes (*Meloidogyne* spp.) (Bellotti et al. 1999). Most of these species target the foliage and stem of the host plant, thereby indirectly impairing the source sink transport, resulting in suppressed root yields (Ceballos et al. 2004). Mites are one of the major pests that cause more significant damage than generalist defoliators. Approximately 50 species of mites have been reported on cassava, consisting of generalist feeders like *Tetranychus* spp. and specialist feeders like *Mononychellus* spp. (Graziosi et al. 2016, Ovalle et al. 2020). In fact, a complex of cassava mites has been reported to cause a yield loss of 15 to 73% to cassava (Byrne et al. 1982, Noerwijati et al. 2022). Until recently, mites were considered a minor pest of cassava; however, mites are now recognized as primary pests of the crop when not adequately managed. Strategies and tactics for management of these pests must be developed for implementation as effective pest management programs for cassava production. To that end, we have reviewed readily available scientific literature on bionomics of the cassava mite complex as well as potential methods of management.

Effective pest management option depends on the agroclimatic condition of the region. Shifting the planting date, appropriate nutrient management, planting resistant or tolerant varieties (Bellotti et al. 2012b) using phytoseiid mites, application of acarofungal pathogens (Bellotti et al. 1994), enforcing quarantine measures to prevent the pests to uninfested areas and preferring selective insecticides for the management of mites are adaptable approaches to check the population of mites (Bellotti et al. 2012b).

Biology and Ecology

Among the mites feeding on cassava, the more important species are cassava green mite (*Mononychellus tanajoa* [Bondar] [syn =*M. progresivus* Doreste]), *M. caribbeanae* McGregor, and *Tetranychus urticae* Koch (syn = *T. cinnabarinus* [Poisd.]), all of which are members of the family Tetranychidae (Acari) (Bellotti et al. 2012b). Other tetranychid species also reported as attacking cassava include *T. tumidus* Banks, *T. kanzawai* Kishida, *T. bellotti* Flechtmann, *T. neocaledonicus* André, *T. truncatus* Ehara, *Oligonychus biharensis* (Hirst), *O. peruvianus* (McGregor), and *O. gossypii* (Zacher), *Eutetranychus orientalis* (Klein) (Bellotti et al. 2012b). *T. urticae* and other species within the genus *Tetranychus* exhibit a wide host range including many field-grown as well as glasshouse cultivated crops. Several noncrop plants also harbor these species. On the other hand, *Mononychellus* feeds solely on cassava (CABI 2024).

Tetranychid mites typically initiate infestation of host plants on the mature leaves near the plant base. They initially infest the abaxial leaf surface and eventually invade the adaxial surfaces resulting in excessive webbing with candlestick symptom during high and severe infestations. Initial plant damage symptoms begin with chlorosis of the leaves which turn yellowish and eventually into a reddish or rust hue as the infestation intensifies. Defoliation of the lower leaves occurs first followed by the upper leaves, it usually leads to the death of the plant, especially in persistent dry climatic conditions (Bellotti et al. 2012c, Bernardo and Esquevia 1981, Ezenwaka et al. 2018) (Fig. 1). Under the webbing, 5 to 10 mites are found per web where the immature stages feed and develop. On reaching adulthood, the mites abandon the webs and form new colonies (Bellotti et al. 2012c). Under South Indian conditions, mites are spatially distributed in their feeding behavior. *Eutetranychus orientalis* and *O. biharensis* feed on the adaxial surfaces of the leaves, while *T. urticae* and *T. neocaledonicus* feed on abaxial surfaces (Edison 2002, Lal and Pillai 1981).

Specifically, *Oligonychus* mites colonize both the surface of mature leaves and the petioles, particularly on the central and middle parts of the plant (Kaimal 2021, Kaimal and Ramani 2011). Small white spots along the leaf veins and margin, which have yellow to brown dots on the corresponding upper surface, can be noticed followed by the formation of dark brown patches, extensive bleaching and chlorosis, desiccation, and defoliation of the leaves with severe infestations (Bellotti and van Schoonhoven 1978, Kaimal and Ramani 2011). Lower leaves exhibit more pronounced damage than upper leaves (Bellotti and van Schoonhoven 1978).

In the case of the cassava green mite, *M. tanajoa*, the infestation manifests as the leaves develop a mottled yellowish-to-whitish color, stippling on the basal half of the leaves at first, later appearing as “pinpricks,” reduction in size of the leaves, deformation of the leaves, mosaic appearance as in the case of cassava mosaic disease, and defoliation with heavy infestations beginning from the apex of the plant often killing the apical and lateral buds and shoots (Chalwe et al. 2015, Ntui et al. 2023, Yaninek and Herren 1988). In coexisting populations of *M. tanajoa* and

Fig. 1. Healthy cassava leaf (A) and mite-infested cassava leaf (B).

T. urticae, both species avoid interspecific competition through habitat stratification which results in a 91% reduction in photosynthetic area of the leaves (James 1988).

The developmental process of tetranychid mites progresses through egg, larva, protonymph, deutonymph, and adulthood with a brief quiescent period of less than a day after each active stage (Bellotti and van Schoonhoven 1978, Cagle 1949, Dosse 1953) (Fig. 2). An adult female *M. tanajoa* oviposits 90–120 eggs on the abaxial leaf surface along on the sides of the midrib or other veins or within concavities on the leaf (Bernardo and Esquevia 1981, Yaseen and Bennett 1976). Freshly laid eggs are typically round, translucent, and smooth, changing their color to yellow-orange with aging (Sakunwarin et al. 2003). For *T. urticae*, the duration of the egg, larval, protonymphal, and deutonymphal stages ranges 3–4, 1–2, 1–2, and 2–3 d, respectively (Bellotti and van Schoonhoven 1978). Development from egg to adult occurs within 7–11 d under optimal temperature (25–28°C) and relative humidity (60–70%) (Bellotti et al. 2012b). A quiescent stage (approximately 2.4 d) follows each instar, during which the mite anchors itself and molts to the succeeding generation (Bernardo and Esquevia 1981, Boudreaux 1963).

Abiotic (e.g., relative humidity, temperature, rainfall) and biotic factors (e.g., plant type, cellular nutrition, and the abundance of natural enemies) regulate the mite populations and influence their development (Mesa et al. 1987). The development, survival, and reproduction of the mites occur best under a temperature ranging from 24–31°C, although a much more comprehensive range is also favorable for development (Win et al. 2018). Higher temperatures lead to an increased net reproductive rate of the mites, leading to an increase in the intrinsic rate of population growth and a reduction in the population doubling time (Bellotti et al. 2012b, Mesa et al. 1987). Rainfall is a critical limiting factor in that heavy rainfall and wind are detrimental to mites (Edison 2002). Increased relative humidity is associated with higher mortality of mites (Boudreaux 1958). Dry ambient conditions and low elevation enhance development, while a low temperature of about 15°C limits the range of *M. tanajoa* (Doreste 1982). Being dry season pests, mite populations move on to new leaf growth, thus, reducing numbers on the more mature leaves during the dry season. Population growth occurs again on the new growth of leaves following the first rains (Herren and Neuenschwander 1991). The initial population surge is attributed to the production of female mites in large numbers (Bellotti et al. 2008). More susceptible plant varieties lose greater amounts of leaf photosynthetic areas to mite attacks than do more resistant varieties, which translates to higher root yield losses in the susceptible lines (Mutisya et al. 2014).

Identification

Tetranychid mites are morphologically identified based on characters of the setae in females and the aedeagus in males. Molecular identification of spider mite employs primers like CO1, ITS1, ITS 2 (Li et al. 2010). DNA barcoding produces reliable results and comparisons. The 5′-end of the mitochondrial cytochrome c oxidase subunit I (COI) gene carries a ∼650 bp region which can be amplified and PCR is performed using appropriate primers (Ovalle et al. 2020).

The two-spotted spider mite, *T. urticae*, occurs in different color forms. The red form, formerly known as *T. cinnabarinus,* was recently confirmed as a color morph

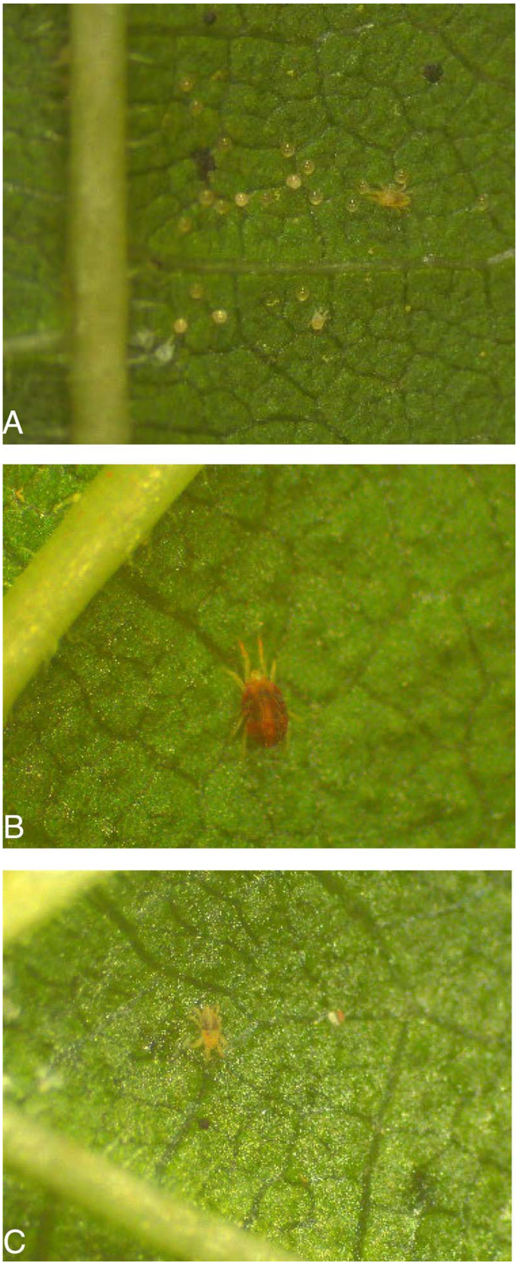

Fig. 2. Tetranychid mite eggs and nymphs (A), adult female (B), and adult male (C).

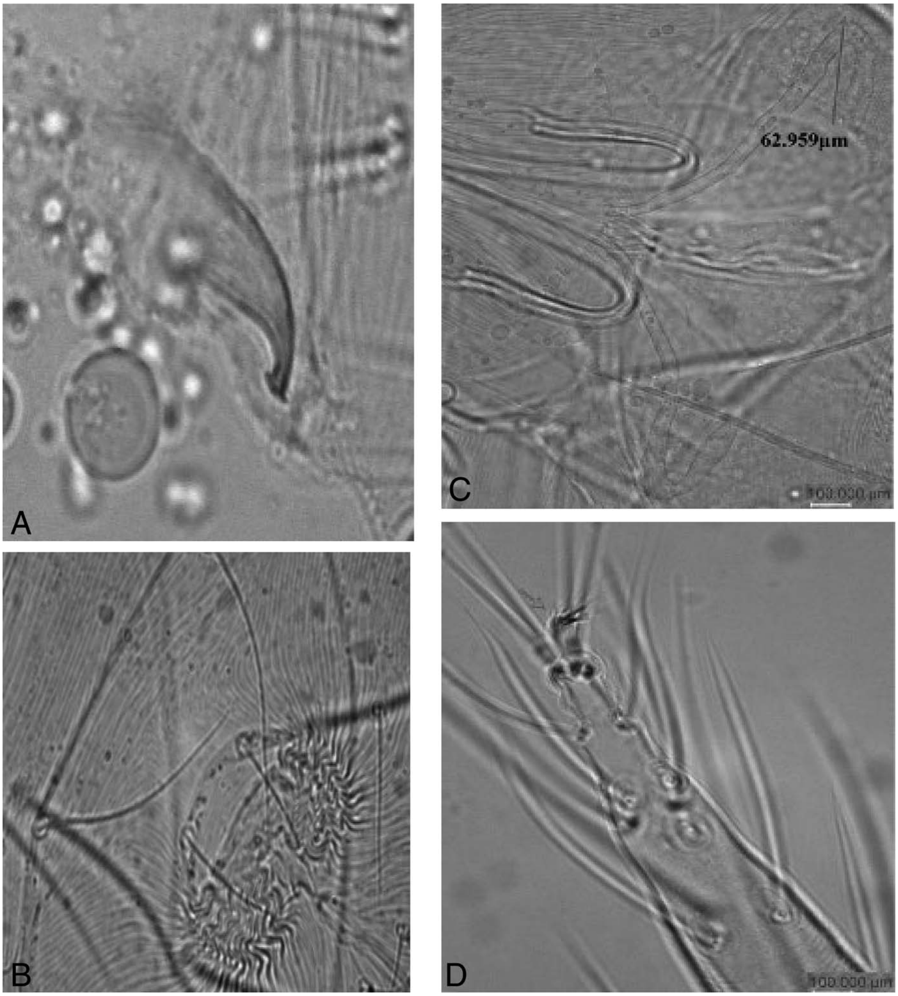

Fig. 3. ***Tetranychus urticae*** **aedeagus (A) female pregenital striae, (B) female peritreme, (C) female and empodium of tarsus I (D).**

of *T. urticae* and is no longer an accepted separate species (Auger et al. 2013). Females are identified by dorsal idiosomal setae that longer than the interval between their longitudinal bases and empodia with 6 proximoventral hairs. The male aedeagus knob is small, and its axis forms a small angle with the axis of the shaft. Anterior and posterior projections of the knob are more or less similar. Embodia I and II, each are with an obvious dorsal spur (Srinivasa et al. 2021) (Fig. 3).

The hysterostomal striae of *T. neocaledonicus* females form a diamond-shaped pattern between the third and fourth dorsoventral setae. Lobes on the dorsal striae are of different shapes and sizes but are primarily semicircular; lobes on medioventral striae are short and wide. The male aedeagus is bent dorsally at nearly a right angle

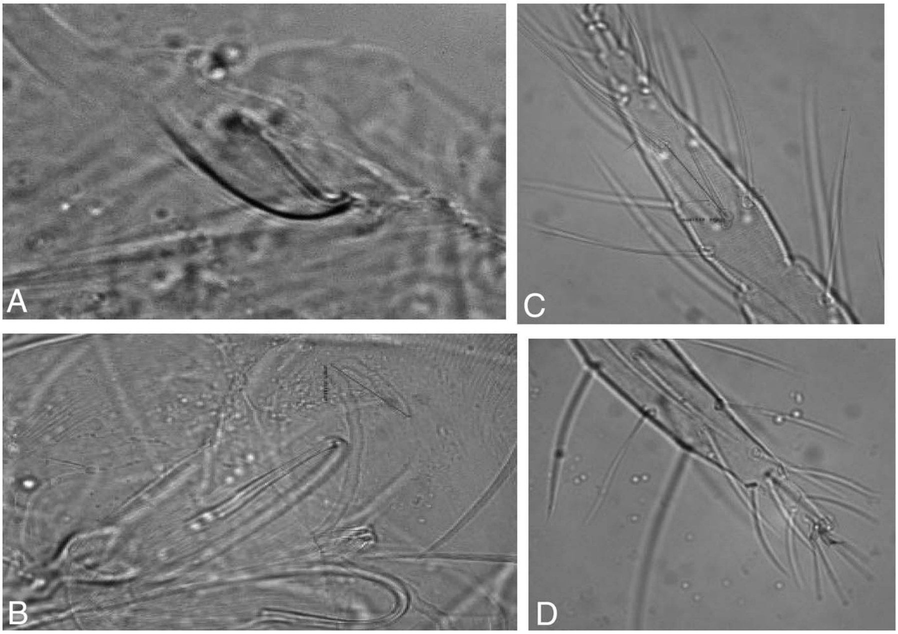

Fig. 4. *Tetranychus truncatus* aedeagus (A), peritreme (B), duplex setae (C), and female empodium of tarsus I (D).

and anterior and posterior projections of the knob are rounded and separated by a small dorsal indentation. Tarsus 1 and 4 have tactile setae and 2 solenidia proximad to proximal duplex setae (Srinivasa et al. 2021).

Tetranychus truncatus (Fig. 4) is polyphagous, feeding on over 86 species of plants distributed in Asia and Southeast Asia (Ehara and Wongsiri 1975, Migeon et al. 2010, Pang et al. 2004, Sakunwarin et al. 2003). Female idosomal setae are longer than the interval between their longitudinal bases and gradually taper distally. Diamond-shaped patterns of striae occur between e1 and f1 setal bases. Tarsus 1 and 4 tactile setae are proximad to proximal duplex setae. The male aedeagus has a tiny terminal knob with an apparent slight indentation on the dorsal margin (Srinivasa et al. 2021).

Mononychellus tanajoa only feeds on cassava (Bellotti et al. 2012c) and is the most serious pest of cassava among the mite species that attack the crop (Poubom et al. 2005). Females are identified taxonomically by the slightly transverse and reticulate pattern of the dorsal striae of hysterosoma and the absence of dorsal body setae on tubercles. The aedeagus of the male possesses distal angulations with a slight apical turn ventrally (Bolland et al. 1998, Ovalle et al. 2020).

Mononychellus caribbeanae is identified by the presence of anastomosed dorsal striae of hysterosoma except in the posterior portion where there is a reticulate pattern in the female. In males, the aedeagal knob has a convex anterior margin with the dorsal and ventral margins on acute angles approximately equal (Bolland et al. 1998, Flechtmann and De Queiroz 2015).

Mononychellus mcgregori (Flechtmann & Baker) is identified by the presence of transverse dorsal striae of hysterosoma and tubercles with dorsal body setae in females. The male aedeagus is with a ventral bend, small triangular knob, rounded posterior margins, and sharp anterior margins (Bolland et al. 1998, Flechtmann and De Queiroz 2015).

Oligonychus biharensis females are characterized with a short and broad sensillum, opisthomal striae transverse, and dorsal idiosoma setae which are thin, simple, slender, and 1.5× longer than the interval between their longitudinal bases. In males, the shaft of the aedeagus is bent dorsad, forming a long knob distally. The knob is longer than the dorsal margin of the shaft, with a small and acute anterior projection and posterior projection that is long, convex, and curved downwards (Srinivasa et al. 2021).

In *Eutetranychus orientalis*, the female sensillum is 4× as long as wide, the dorsal sensillum conical, and about half of the terminal sensillum. Dorsal idiosomal setae are borne on strong tubercles and are hairy. Striations occur on the propodosomal region and longitudinal and hysterosomal striations sometimes form a triangular pattern. Males have a terminal sensillum of palpus 3× as long as wide, and the dorsal sensillum is long and slender. The aedeagus is more or less a hooked-like structure with a distal bent portion longer than the dorsal margin of the shaft, which is slightly concave (Srinivasa et al. 2021).

Distribution and Range

Native to South America, *M. tanajoa* was accidentally introduced into Uganda in the early 1970s, spreading rapidly extending several hundred km per year to 27 countries of the cassava belt by 1985, causing estimated root yield losses of 13 to 80% (Bellotti et al. 2012b, Herren and Neuenschwander 1991, Nukenine et al. 1999, Yaninek and Herren 1988). *Tetranychus urticae* is distributed throughout the cassava-growing regions of the world and primarily in Asia (Zou et al. 2018). *Oligonychus peruvianus* occurs only in the Americas, but *O. gossypii* has been reported from Africa. While *T. urticae* has a host range of a wide variety of crops, both *M. tanajoa* and *O. peruvianus* are limited to primarily *Manihot* spp. (Bellotti and van Schoonhoven 1978).

The tropical climate of several cassava-growing Asian countries is highly conducive to the rapid dispersal and outbreaks of *M. tanajoa* (Bellotti et al. 2008). The local spread of cassava mites is through wind, while dispersal and spread across greater distances is usually attributed to movement of infested cassava stem cuttings (Yaninek et al. 1989). Interchange and replanting of the mite-infested plant materials contributes to the spread of the mite pests (James 1988). An increase in the movement of people, equipment, and setts, along with factors such as the changing climate, may contribute to the spread of mites (Smith et al. 2018). Modern transport facilities have aided in the movement of neotropical pests to Africa.

Overview of Economic Loss

Cassava yield losses ranging from 21 to 80% have been reported and appear dependent on several factors such as location or region, plant varietal susceptibility, and period and duration of the attack and infestation (Bellotti et al. 2012a). Under field conditions, yield losses of 2 to 10% in reports of slight damage up to 60% in reports of severe damage have been reported from Asia (Ovalle et al. 2020). In experimental trials, fresh yield losses recorded in susceptible cultivars at 3, 4, and 6 mo after planting were 21, 25, and 53%, respectively. Resistant cultivars have exhibited yield losses of 15%, while other varieties experience 67% yield loss under similar conditions (Bellotti et al. 2012c, Herrera et al. 2011).

Some differences in losses have also been observed with different mite species. *Tetranychus urticae* infestations reportedly caused a 50 to 70% decline in cassava production (Lu et al. 2017). It also caused a yield reduction of 50% in Indonesia (Graziosi et al. 2016), and when mixed with infestations with *M. tanajoa*, caused yield losses as high as 53% (Bellotti and Byrne 1979). Furthermore, infestation by a complex of mites (*T. urticae, T. cinnabarinus, M. caribbeanae, M. tanajoa,* and *M. progressivus*) lowered root yield by 87% and the number of stem cuttings by 82% (Ovalle et al. 2020). Field trials have reported yield losses of 18 to 47% due to *T. kanzawai* in the Philippines, *E. orientalis, T. neocaledonicus* and *O. biharensis* in India, and *Tetranychus* sp. in Indonesia (Bellotti et al. 2012b). Field studies at the International Center for Tropical Agriculture (CIAT) found that a complex of mite species (*M. tanajoa, M. mcgregori, T. urticae, and O. peruvianus*) resulted in 20 to 53% losses (Bellotti and van Schoonhoven 1978). *Mononychellus mcgregori* also caused 60% reduction in yield in China in a study involving population dynamics, host plant resistance, and the time of the mite attack (Chen et al. 2010, Lu et al. 2014).

The cassava green mite, *M. tanajoa*, has caused severe yield losses ranging from 21 to 53% on commercial plantations (Bellotti et al. 2012a) and 30% in field experiments (absence of natural enemies) conducted in Colombia (Braun et al. 1989), 46% in experimental plots in Uganda and 15 to 20% in Venezuela (Bellotti and van Schoonhoven 1978), and 10 to 80% in trials in Africa (Robert et al. 2016). Infestations have also caused 60% reduction in leaf biomass (Mutisya et al. 2015b), 30 to 50% reduction in fresh storage root in Zambia (Chalwe et al. 2015), a 29% decline in the dry matter of storage roots and a 21% reduction in dry matter of stems in field trials in Kenya (Skovgård et al. 1993).

In all, mite infestations reduce the quality of the planting material for the upcoming growing season and indirectly by increasing the price of planting material (Robert et al. 2016, Walker 1987). Yield losses often indirectly affect the price of the commodity, other alternatives, and food imports (Walker 1987).

Management of the Cassava Mite Complex

Historically, traditional cassava plantations in which mites were not a concern because the occurrence of natural enemies kept mite populations below crop damaging levels (Yaninek et al. 1989). Commercial monocultures of the crop, however, rely increasingly on broad-spectrum chemical pesticides for pest management. This practice reduces or eliminates populations of beneficial natural enemies leading to the elevation of mite species from a secondary pest status to primary pest status (Bellotti and van Schoonhoven 1978, Bellotti et al. 2012c). Indiscriminate use of these conventional chemistries also can lead to the development of resistance as was reported with *T. africanus* Tucker populations in Thailand (Graziosi et al. 2016). Chemical control methods are generally not available for most African farmers (Nyiira 1976, Yaseen and Bennett 1976), and their use in managing mites is not economically feasible for low-income farmers globally.

Cultural control. A few cultural management methods offer limited relief (Bellotti et al. 1994). Adjusting planting dates, removal of mite-infested leaves, timely planting at the beginning of the rainy season (Yaninek et al. 1989), and nutrient management through increased application of nitrogen and phosphorus fertilizers suppresses the mite populations (Lal and Pillai 1981). Resistant germplasm (Herren 1994), biocontrol agents, genetic improvement of natural enemies, fungal pathogens, augmentation and conservation of natural enemies, and habitat manipulation should be thoroughly explored for management of these phytophagous mites (Bellotti et al. 1994).

Host plant resistance (HPR). Utilizing pest-resistant plants is an environmentally friendly and cost-effective means for cassava pest management (Indiati et al. 2023). CIAT, IITA, and EMBRAPA (The Brazilian Agricultural Research Corporation) conduct research to incorporate resistance into cassava cultivars (Bellotti et al. 2008, Nassar and Ortiz 2007). Host resistance to mite pests is absent in cassava; only low-to-moderate resistance (6% of 5000 plants screened) against *M. tanajoa* is found in the CIAT germplasm bank. Development of moderately resistant plants can suppress the initial mite population growth, providing an opportunity to effectively use natural enemies as a management tactic (Bellotti and van Schoonhoven 1978, Bellotti et al. 2012c). In fact, preliminary screening at CIAT indicated a greater resistance against *M. tanajoa* (controlled by additive gene effects) and *O. peruvianus* compared to *T. urticae* (Bellotti and Byrne 1979, Bellotti et al. 1987).

In general, mites feeding on susceptible plants exhibited high fecundity and development rates, easy acceptance of the plants, long adult life span, and low nymphal mortality rates in comparison to high mortality rates, long development periods, and short oviposition periods for those fed on resistant plants (Bellotti et al. 2012b). The mechanism of *Mononychellus* mite resistance is based on altering mite biology, plant surface pubescence, and plant tolerance to mite attack (Bellotti et al. 1987), while resistance to *T. uticae* is based on leaf color and wax layer thickness (Wei et al. 2009). Genotypes with high intra- and inter-season stability of traits conferring resistance to the cassava green mite will ensure more durable HPR (Able et al. 2023). Peroxidases and polyphenoloxidases can play a vital role in increasing the resistance of cassava to mites (Liang et al. 2017).

Screening of CIAT germplasm yielded seventeen cultivars resistant to *T. urticae* and *Mononychellus*. Screening by Visayas State College of Agriculture, Philippines showed 4 cultivars resistant to *T. kanzawai* (Bellotti et al. 1987). *Manihot flabellifolia* Pohl (Malpighiales: Euphorbiaceae) and *Manihot peruviana* Müll. Arg (Malpighiales: Euphorbiaceae) exhibited high levels of resistance against *M. tanajoa.* Cassava varieties like M Bra 12, M Col 1434, and M Ven 125 are resistant sources against the pest (Parsa et al. 2015). Cassava accessions from CTCRI, including CE-4, CE-14, CE-38, and CE-139, were highly resistant to spider mites (Edison 2002). A greenhouse study at the Indonesian Legumes and Tubers Crop Research Institute found that 4 clones (CMM 03036-7, CMM 03036-5, CMM 03038-7, and CMM 02040-1) and 1 cultivar (Adira 4) were moderately resistant to cassava mites while the clones CMM 03036-7, CMM 03036-5, CMM 03038-7, and CMM 02040-1 were found to be tolerant to mite feeding (Indiati et al. 2023). Eight cultivars (e.g., Adira 1, Adira 2, Adira 4, Malang 1, Malang 4, Malang 6, LITBANG UK-2, and UK-1 Agritan) were reported to be resistant to red spider mite (Indiati et al. 2023). Greenhouse studied in China revealed that C1115 and MIANDIAN were highly resistant and SC5, SC9, SC15, COLUMBIA-4D and LIMIN were resistant varieties against *T. urticae* (Liang et al. 2022). The Central Tuber Crop Research Institute (Thiruvananthapuram) developed varieties H-97 (resistant), H-165, H-226, Sree Harsha, Sree Rekha, and Sree Prabha (tolerant), and Sree Visakham (less susceptible) to cassava red spider mite (CTCRI 2023).

Biological control. Biological control of mite pests can be through classical biological control, conservation, or augmentation, which could offer a long-lasting solution for mite pest control (Herren 1994). Around 250 species of natural enemies, including parasitoids, predators, and pathogens, are documented as attacking pests of cassava. In general, populations of predators are density dependent and, thus, increase as the mite populations increase during the dry season and decrease when there is a decline in the mite population during the rainy period (Bellotti and van Schoonhoven 1978). The common predator families that feed on the cassava mites are listed in Table 1.

When the cassava green mite was initially identified as a pest of cassava, there were no effective control measures against the pest because of lack of finances to afford support management programs and a general neglect of the management of the cassava crop (Yaninek and Herren 1988). The field population of natural enemies kept mite populations below economically damaging levels in the Neotropics (Yaseen 1986); however, the indigenous natural enemies in Africa were ineffective against the exotic cassava green mite when accidentally introduced there. Attempts with classical biocontrol of the cassava green mite in Africa were coordinated by the Commonwealth Institute of Biological Control (CIBC) (Markham et al. 1987). *Orthotylus minutus* Jakovlev (Hemiptera: Miridae) and 2 species of phytoseiid mites were imported from Trinidad, but the program was not successful due to the lack of proper infrastructure facilities and trained personnel (Murphy 1985). Later, the International Institute of Tropical Agriculture (IITA) launched the African Biological Control Programme (ABCP) in 1988 (Yaninek et al. 1989). The biological control methods for management under this program began in Africa in the 1980s when the Neotropics were explored (2,400 sites of 14 countries) for natural enemies. Because little was known about the natural enemies, agroecological conditions were characterized, followed by matching with agrometeorological data, leading to the choice of phytoseiid mites, owing to their similarity with the spider mite in terms of ecology and occurrence (Toko et al. 2019, Yaninek and Herren 1988). More than 10 species of phytoseiid mites were released between 1983–1988, and more than 11 million individual predators were released between 1984–2001 (Yaninek and Hanna 2003). Many of the mite predators, such as *O. minutus Cameron* (Coleoptera: Staphylinidae), *Stethorus sp.* (Coleoptera: Coccinellidae), *Scolothrips* sp. (Thysanoptera: Thripidae), *Anthrocnodax* sp. (Diptera: Cecidomyiidae), were found to be effective only at higher mite populations (Yaninek et al. 1989).

Indeed, phytoseiid mites appear to possess great potential as effective predators for controlling phytophagous mites as they are specific and effective at low mite populations (Huffaker et al. 1970). The phytoseiid predator *Neoseiulus anonymus* Chant

Table 1. Biological control agents of cassava mites.

Biological Control Agent	Species	Reference
Predatory mites	*Euseius fustis* (Pritchard and Baker)	Markham et al. 1987
	E. lokele (Pritchard and Baker)	Markham et al. 1987
	E. talinga (Pritchard and Baker)	Markham et al. 1987
	E. papayan (Pritchard and Baker)	Markham et al. 1987
	E. baetae (Meyer and Rodrigues)	Bellotti et al. 2012b
	E. concordis (Chant)	Bellotti et al. 2012b
	E. ncholsi Ehara and Lee	Bellotti et al. 2012b
	Galendromus annectens (De Leon)	Bellotti et al. 2012b
	G. helveolus (Chant)	Herren and Neuenschwander 1991
	Neoseiulus idaeus Denmark and Muma	Bellotti et al. 2012b
	N. californicus (McGregor)	Herren and Neuenschwander 1991
	N. anonymous (Chant and Baker)	Bellotti et al. 2012b
	N. teke (Pritchard and Baker)	Markham et al. 1987
	Phytoseiulus macropilis (Banks)	Bellotti et al. 2012b
	Typhlodromalus aripo De Leon	Bellotti et al. 2012b
	T. manihoti Moraes	Bellotti et al. 2012b
	T. limonicus (Garman and McGregor)	Bellotti and van Schoonhoven 1978
	T. rapax De Leon	Yaseen and Bennett 1976
	T. saltus (Denmark and Matthysse)	Markham et al. 1987
Predatory insects		
Cecidomyiidae	*Therodiplosis persicae* Kieffer	Yaseen and Bennett 1976
	Arthrocnodax sp.	Chazeau 1985
Staphylinidae	*Oligota minuta* Cameron	Chazeau 1985
	O. barbadorum Frank	Yaseen and Bennett 1976
	O. fageli Williams	Chazeau 1985
	O. pallidicornis Cameron	Chazeau 1985
	O. pygmaea (Solier)	Chazeau 1985
Phalacridae	*Phalacrus sp.*	Markham et al. 1987
	Stilbus sp.	Markham et al. 1987
Latridiidae	*Melanophthalma sp.*	Markham et al. 1987
Corylophidae	*Arthrolips sp.*	Markham et al. 1987
Coccinellidae	*Stethorus aethiops Wse*	Markham et al. 1987
	S. jejunus Casey	Markham et al. 1987
	S. darwini (Bréthes)	Chazeau 1985
	S. exsultabili Chazeau	Chazeau 1985
	Cheilomenes sp.	Bellotti and van Schoonhoven 1978
	Chilocorus distigma Klug	Markham et al. 1987
	Hyperaspis pumila Mulsant	Markham et al. 1987
	Micraspis striata (Fabricius)	Markham et al. 1987
	Brumoides nigrifrons (Gerstaecker)	Markham et al. 1987
	Scymnus morelleti Mulsant, *S. scapuliferus* Mulsant	Markham et al. 1987

Table 1. Continued.

Biological Control Agent	Species	Reference
	Cheilomenes sulphured (Olivier)	Markham et al. 1987
	Exochomus ventralis Gerstaecker, *Exochomus sp*.	Markham et al. 1987
Anthocoridae	*Verania sp.*	Bellotti and van Schoonhoven 1978
	Orius insidious (Say)	Bellotti and van Schoonhoven 1978
	Clamidiastethus sp.	Chazeau 1985
	Wollastoniella gatti Ghauri	Markham et al. 1987
	Xylocoris sp.	Markham et al. 1987
Lygaeidae	*Geocoris sp.*	Chazeau 1985
Thysanoptera	*Scolothrips pallidus* (Beach)	Chazeau 1985
	Frankliniella sp.	Markham et al. 1987
Neuroptera	*Coniopteryx crassicornis* Eb. Pet.	Chazeau 1985
	Conwentiza Africana Meinander	Markham et al. 1987
Fungus	*Neozygites floridana* (Weiser and Muma)	Bellotti et al. 2012b
	Hirsutella thompsonii Fisher	Bellotti et al. 2012b
	Entomophthora sp.	Herren and Neuenschwander 1991

and Baker (Acari: Phytoseiidae) is short-lived, completing its life cycle within half the time taken by cassava green mite and *Tetranychus* mites (Bellotti et al. 2008). Field studies in Colombia demonstrated the efficacy of biological control using these predators over the use of chemical acaricides. The acaricide treatments reduced natural enemy populations resulting in a yield loss of up to 33%, while the biological control method alone prevented yield loss (Braun et al. 1989).

In the late 1990s, the predatory phytoseiid mites *T. aripo* De Leon (Acari: Phytoseiidae) and *T. manihoti* (Moraes) (Acari: Phytoseiidae) were successfully introduced and established along with a fungal pathogen *Neozygites tanajoae* Delalibera Jr., Humber & Hajek (Zygomycetes: Entomophthorales) by IITA personnel in West Africa. These natural enemies originated from areas in Brazil with similar climatic conditions (Delalibera 2002, Ortiz 2006) Within 2 yr, the predatory mites had reduced the cassava green mite densities by 30 to 60% with a concomitant 15 to 35% increase in cassava yields, and epizootics of *N. tanajoae* further reduced the cassava green mite population by 25% (Ortiz 2006). This cassava green mite biological control program was a success as it achieved a 100-fold economic benefit (Dixon et al. 2003).

The phytoseiids *E. ho* De Leon, *T. aripo*, *T. tenuiscutus* McMurtry & Moraes, *N. californicus* (McGregor), *N. idaeus* Denmark & Muma, and *G. annectens* De Leon were tested against *M. tanajoa,* with *E. ho*, *T. aripo*, and *T. tenuiscutus* being determined as the more efficacious of the six predators (Cuellar et al. 2001). *Typhlodromalus aripo* reduced the cassava green mite population up to 60%, which increased cassava fresh matter production by up to 37% (Bellotti et al. 2012c). In Kenya, *T. aripo* reduced the intensity of *M. progresivus* in cassava. The release of a phytoseiid predator, *N. idaeus*, in East Africa had no impact on the mite populations (Markham et al. 1987). The phytoseiid mite *Neoseiulus (Amblyseius) longispinosus* (Evans) has reportedly controlled *T. kanzawai, T. truncatus, T. urticae,* and *E. africanus* in cassava in the Philippines and Thailand (Graziosi et al. 2016).

Mutisya (2014) reported that factors like the initial ratio of predator to prey and the diet preference of different species impact management of phytophagous mites by predatory mites. Wild Manihot species, such as *M. tristi* Müll. Arg, *M. filamentosa* Pittier, and *M. alutacea* D.J. Rogers & Appan, carry resistant genes and, hence, reduce the oviposition by *M. tanajoa* when compared to a susceptible *M. esculenta* (CMC 40) cultivar. The phytoseiids *G. annecteus*, *E. concordis* (Chant), *Phytoseiulus persimilis* Athias-Henriot, *N. anonymous*, *N. idaeus*, and *P. macropilis* (Banks) feed on *Tetranychus* mites with *N. anonymous* feeding on all developmental stages of *T. urticae* (Mesa et al. 1990). Phytoseiids are highly sensitive to even mild disturbances, as was evident from the neotropics when pesticides used against thrips in cassava caused mortality of phytoseiids resulting in an increased phytophagous mite population with in yield losses (Braun et al. 1989). Field studied in West Africa using *T. aripo*, *Amblydromalus manihoti* (Moraes) (Acari: Phytoseiidae), and *E. fustis* suggested that *T. aripo* is the more effective predator among the three, and *T. aripo* along with *E. fustis* could only reduce the population of *O. gossypi* in cassava (Onzo et al. 2014).

Limiting factors for implementation of biocontrol efforts include the high cost of initiating the program and incompatibility with other control methods. Abamectin, chlorpyriphos, and the predator *T. aripo* were used to manage *M. progresivus* in Kenya. In that study, abamectin was effective in controlling the pest under all agro-ecological conditions. At the same time, *T. aripo* helped suppress the population of the mites by 45% in the dry hot midlands, by 64% in the warm humid coastal climatic zones, but it did not survive in the cool midlands. However, high rainfall and the presence of the phytoseiid *E. fustis* helped in the reduction of mite populations in the cool midlands (Mutisya et al. 2015a).

Other promising predators for cassava green mite management include the rove beetle *Oligota minuta* Cameron (Coleoptera: Staphylinidae) and the coccinellid *Stethorus* sp. (Coleoptera: Coccinellidae) (Bellotti et al. 2012c). IITA rears and releases phytoseiids like *Amblyseius aerialis* (Muma)*, E. concordis, N. anonymus, N. californicus, N. idaeus, T. limonicus, G. annectens,* and *G. helveolus* in various programs. The mites are fed *T. urticae* on leguminous plants and fed with *T. urticae* (Herren and Neuenschwander 1991, Yaninek and Herren 1988) and could prove useful for cassava mite complex management.

As referenced earlier herein, the fungal pathogen *N. tanajoae* is an effective fungal pathogen specific to *M. tanajoa* in Africa (Delalibera et al. 2004). *Neozygites cf floridana* (Weiser and Muma) (Entomophthorales: Neozygitaceae) also infects *M. tanajoa* in Brazil (Bellotti et al. 1999, Toko et al. 2019). It survives as resting spores during the dry season on other hosts. With the onset of the wet season, the fungus can be found on cassava (Elliot et al. 2000), where It caused mortality of the cassava green mite within 1–2 wk (Bellotti et al. 2008). The epizootics by *N. floridana* caused higher relative mortality in the adult female stages compared with other stages of the mites (Elliot et al. 2002). Utilizing *N. floridana*, along with the influence of other factors like lower temperature, reduced food quality, and other environmental factors influencing fungal development, can help check the mites populations (Alonso et al. 2021). *Neozygites floridana* caused 10% mortality of the *M. tanajoa* population and 19% of the *O. gossypii* population, while *Hirsutella thompsonii* Fisher killed 19% of the mites (Yaninek et al. 1996). *Hirsutella* sp. and *Entomophthora* sp. also have been recorded infecting the cassava green mite (Herren and Neuenschwander 1991). *Hirsutella thompsonii* is a promising biocontrol agent against cassava green mite when it is applied during weather conditions conducive to the fungus, for example, rainfall (Odongo et al. 1998). *Beauveria bassiana* (Balsama-Criv.) Vuillemin (Hypocreales: Cordycipitaceae) and *Metarhizium anisopliae* (Metschinkoff) Sorokin (Hypocreales: Clavicipitaceae) isolates at a concentration of 5×10^7 conidia/ml caused >80% and 90% mortality, respectively, while *Hirsutella* sp. at a concentration of 1.7×10^7 conidia/ml caused 73% mortality *T. urticae* (Maniania et al. 2009). *In vitro* assessment of *B. bassiana* caused up to 97% mortality of *T. truncatus* (Chaithra et al. 2022) and 13 to 97% mortality of cassava green mite, while *M. anisopliae* caused 12 to 45% mortality of cassava green mite (Barreto et al. 2004).

Botanicals. The use of botanicals is an acceptable alternative to conventional chemical acaricides (Janlaor and Auamcharoen 2021). Neem oil is a selective botanical that can effectively reduce fecundity and cause mortality in the immature and adult stages of cassava green mite (Silva et al. 2013). The essential oils extracted from dill (*Anethum graveolens* L. [Apiales: Apiaceae]) and garlic (*Allium sativum* L. [Asparagales: Amaryllidaceae]) when tested against *T. urticae* and *T. truncatus* found that 20% (v/v) *A. sativum* oil caused 73.5% mortality of *T. truncatus* adult females as a contact toxin. In testing, *A. graveolens* oil at 5 to 20% was highly repellent to 98 to 100%, 62 to 96%, and 86 to 100% repellent to *T. urticae* (86–100%) and 98 to 100% repellent against *T. truncatus* (Sararit and Auamcharoen 2020). *Helicteres angustifolia* L. (Malvales: Malvaceae) extract repelled 97.78% of adult *O. biharensis.* That extract also reduced oviposition by *Melodinus suaveolens* Champ. (Gentianales: Apocyanaceae), *Pterospermum heterophyllum* Hans (Malvales: Malvaceae), *H. angustifolia*, and *Datura metel* L. (Solanales: Solanaceae) by 100% (Fangping et al. 2006). The acetone extract of *Aloe vera* (L.) Burman against *T. cinnabarinus* functioned as an effective botanical acaricide with a median lethal concentration (LC_{50}) of 90 ppm (Wei et al. 2011). Essential oils extracted from the fresh and dried rhizomes of the *Acorus calamus* L. (Acorales: Acoraceae) caused a mortality levels in *T. truncatus* adults of 73.8% with the fresh rhizomes and 91.8% with the dried rhizomes. Direct contact toxicity assays using essential oils from fresh rhizomes reduced the egg hatch by 96.3% when applied at a 5% (v/v) concentration (Janlaor and Auamcharoen 2021). Cassava green mite subjected to concentrations equivalent to the LC_{25} or LC_{50} of acid lime essential oil reduced mortality while inducing a high predatory response by a generalist predator, *Ceraeochrysa caligata* (Banks) (Neuroptera: Chrysopidae) soon after the exposure (Farias et al. 2020). Application of methyl jasmonate to cassava induced defense responses in cassava towards *T. urticae* and reduced egg production and adult longevity of the mite with slower development and extension of the duration of egg development of the mites (Zhang et al. 2023).

Conventional chemical acaricides. The use of acaricides to control mites on cassava cannot be justified for the warm wet and cool wet areas as high populations of predatory mites occur naturally in both conditions (Kariuki et al. 2005) and high rainfall in the warm wet areas maintain mite populations under economic levels (Yaninek et al. 1989).

Emamectin benzoate and abamectin at concentrations of 100 mg/L and 0.1 mg/L caused 100% mortality of *T. kanzawai* female adults. Furthermore, abamectin at this concentration proved safe to *Amblyseius longispinosus* (Evans), but emamectin at those concentrations was fatal to the predator (Villacencio and Vasquez 2022). Out of 27 pesticides tested against mite complexes, ethion (0.05%), dimethoate (0.05%), monocrotophos (0.03%), and dicofol (0.05%) were effective (Lal and Pillai 1981).

Development of resistance to acaricides becomes a major challenge in mite management programs (Sato et al. 2016). Due to its short life cycle, arrhenotokous reproduction, inbreeding, and high fecundity, *T. urticae* has reportedly developed resistance to several organophosphates, carbamates, synthetic pyrethroids, mitochondrial electron transport inhibitors (METIs), and others (Van Leeuwen et al. 2010). Resistance to spiromesifen and fenpyroximate also has been reported (Sato et al. 2004, 2016). Resistance of the *T. truncatus* strain VkOk1 to spiromesifen, diafenthiuron, and fenazaquin is reportedly 8-, 10-, and 13-fold greater than nonresistant strains in Kerala (Bachhar et al. 2019).

Concluding Remarks

Because cassava plays a vital role as a staple food and a raw material for industrial uses, efforts must be made to protect the crop from diseases and other pests. Of great importance is the complex of mite pests that have caused severe economic losses to cassava throughout areas in which it is grown and produced globally. Proper identification and management of the mites are essential. Different management strategies can be incorporated in the integrated pest management (IPM), taking into account the agroclimatic conditions of the area where the pest is limiting cassava production. Studies must be initiated and conducted for the sustainable management of the pest. This review will hopefully provide a foundation for these future efforts.

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