Life Cycle and Larval Morphology of *Diomus terminatus* (Coleoptera: Coccinellidae) and Its Potential as a Biological Control Agent of *Melanaphis sacchari* (Hemiptera: Aphididae)

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ABSTRACT The life cycle, morphology, and potential of *Diomus terminatus* Say (Coleoptera: Coccinellidae) to control the sugarcane aphid, *Melanaphis sacchari* (Zehntner) (Hemiptera: Aphididae), were examined in the laboratory. The morphologies of first and fourth instars are described and illustrated, with emphasis on characters of first instar that differ from those of later instars. Dramatic differences exist in the relative size and abundance of specialized setae between early and late instars. Field-collected late instars of *D. terminatus* were successfully reared to adult stage at 26°C by using *M. sacchari* feeding on small pieces of sugarcane (*Saccharum* spp.) leaves. The eggs laid on the sugarcane leaves hatched in 4.5 ± 0.1 d. Larval and pupal stages lasted an average of 6.8 ± 0.6 and 4.9 ± 0.2 d, respectively. *D. terminatus* required 12.1 ± 0.6 d from egg hatch to adult emergence. The adult longevity test with single adults in petri dishes revealed that *D. terminatus* remained alive for 26 ± 1.9 d when feeding on aphids of mixed ages. The larvae consumed a total of 30 ± 1.8 aphid nymphs, with a daily consumption rate of 4.7 ± 0.4. The adult voracity test showed that *D. terminatus* could consume as many as 19 ± 0.9 aphids per d. These results are discussed with respect to their implications for aphid control in Louisiana sugarcane.

KEY WORDS *Diomus terminatus*, *Melanaphis sacchari*, life cycle, morphology, biological control

The sugarcane aphid, *Melanaphis sacchari* (Zehntner) (Hemiptera: Aphididae), a small ant-tended hemipteran with various body colors, is distributed throughout the tropical and subtropical regions (Blackman and Eastop 1984). The first finding in Louisiana was reported on 9 September 1999, on the USDA–ARS Ardoyne Research Farm near Houma, and a subsequent survey showed that eight out of 21 parishes where sugarcane is planted were infested (White et al. 2001). *M. sacchari* is a key pest of sorghum (*Sorghum* spp.) and of sugarcane in many parts of Africa, Asia, Australia, the Far East, and in Central and South America (Singh et al. 2004). Other hosts include rice (*Oryza sativa* L.), maize (*Zea mays* L.), millet (*Setaria* spp.), barnyard grass (*Panicum colonum* L.), bermuda grass [*Cynodon dactylon* (L.) Pers.], and several additional grasses. Feeding by *M. sacchari* on sugarcane leaves causes a slight loss of leaf greenness, and heavily infested leaves look black with excessive production of honeydew, leading to the development of sooty mold (Hall and Bennett 1994). *M. sacchari* is also an important vector of sugarcane yellow leaf virus especially in Hawaii where the infection level in several commercial cultivars reached up to 95% (Schenck and Lehrer 2000). Recent studies in Louisiana indicated that *M. sacchari* was the most abundant aphid species recorded in biweekly surveys, and up to 25% of the area within fields in several locations was infected with yellow leaf virus disease (McAllister et al. 2005). Sugar yield losses up to 11 and 14% have been reported in first and second ratoon crops, respectively, in Louisiana because of the sugarcane yellow leaf virus (Grisham et al. 2001). To minimize the spread of virus, yellow leaf has been added to the certification standards for micropropagated seedcane.

Singh et al. (2004) presented a comprehensive review of *M. sacchari* biology and listed >47 natural enemies in different countries. These included pathogens (*Verticillium lecanii*), parasitoids (Hymenoptera), and predators (Diptera, Neuroptera, Coleoptera, and Hemiptera). Among these groups, lady beetles (Coccinellidae), lacewings (Chrysopidae), and hover flies (Syrphidae) seemed more important because they cause greatest mortality to the *M. sacchari* populations (Singh et al. 2004).

Diomus terminatus (Say) (Coleoptera: Coccinellidae) is a generalist aphid predator native to the eastern and midwestern United States (Gordon 1976). It has been successfully reared under laboratory conditions on several aphids, including the yellow sugarcane aphid, *Sipha flava* (Forbes) (Hall 2001, Hentz and Nuessly 2002), corn leaf aphid, *Rhopalosiphum maidis* (Fitch), cotton aphid, *Aphis gossypii* Glover, and green peach aphid, *Myzus persicae* (Sulzer) (Hallborg 2003). This species was observed feeding on *M. sacchari* in Louisiana (White et al. 2001), but studies

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have not been conducted on its life cycle by using this aphid as prey. Larvae of this beetle were noticed feeding on *M. sacchari* in a variety field test near Youngsville, LA, on 10 July 2007. The larvae were collected, reared in the laboratory, and studied for biological control potential. Hentz and Nuessly (2002) provided cursory descriptions of various life stages of *D. terminatus*, but details of taxonomically informative characters were not given. Ślipiński (2007) provided a generic larval description based on the Australian species *Diomus notescens* (Blackburn) and an unidentified *Diomus* sp., including illustrations of the latter. He also discussed issues related to the generic diagnosis of the genus involving adult characters. A detailed morphological description of first and fourth instars of *D. terminatus* is also provided here. These descriptions will allow integration of characters into phylogenetic analyses of coccinellids and other cucujoid beetle taxa, and they provide a more comprehensive basis for distinguishing larvae of this species from those of other coccinellids.

Materials and Methods

Life Cycle Studies. The *D. terminatus* colony was initiated in July 2007 with 25 late instars collected from a small-plot sugarcane (*Saccharum* spp.) variety test near Youngsville (Lafayette Parish, LA). *M. sacchari* feeding on small cut pieces of sugarcane leaves were used as prey in this study. This was important to mimic natural conditions and to avoid loss of any plant physical or chemical cues that might be helpful to *D. terminatus* in finding and using its prey (Hallborg 2003). Beetles were provided with fresh aphids every 1–2 d that were collected either from an aphid colony in the greenhouse or directly from the field at the LSU AgCenter's St. Gabriel Sugar Research Station (Iberville Parish, LA). The beetle colony was maintained in an incubator at 26°C, 75 ± 5% RH, and a photoperiod of 14:10 (L:D) h. The life stage studies were conducted in the same incubator. Beetle larvae along with *M. sacchari* on cut sugarcane leaves were brought into the laboratory and placed in a rearing chamber, a 42,875-cm^3 Plexiglas box with a round opening of 10 cm in diameter covered with perforated plastic to prevent beetle escape and ensure ventilation. Larvae also were provided with a 20% sugar solution as an additional food source inside the chamber, and wax paper and Kimwipes as pupation and oviposition sites for larvae and adults, respectively.

Newly hatched larvae were taken out of the chamber and placed individually in 15-ml scintillation vials to avoid cannibalism or reduced survivorship due to insufficient aphid supply (Hallborg 2003). Each first instar was provided with 10–15 aphid nymphs feeding on three to four ≈5-cm pieces of sugarcane leaf. The later instars were provided with 15–20 nymphs. Vials were examined daily to record exuviae and number of aphids consumed. The old leaf and aphids were replaced with fresh leaf pieces and aphids until the last instar was seen stuck at its posterior end, an indication of initiation of pupation. Larvae were transferred into clean vials when needed. The length and maximum width of pupae, their preferred location for pupation on the leaf piece, day of pupation for each surviving larva, and day of emergence for each surviving pupa were recorded. Adult beetles emerging from each vial were placed in a petri dish (8.5 cm in diameter, at least four beetles per petri dish) with a moist cotton ball and several aphids on three or four ≈5-cm sugarcane leaf pieces. The gender of the beetles was not determined at this stage. However, based on visual determination of differences in body size, the beetles were placed in petri dishes in a target male to female ratio of 1:1. The presence of females in petri dishes was confirmed by observing eggs on leaves or on the bottom of petri dishes the next day. Beetles were transferred into new petri dishes with fresh aphids and sugarcane leaves every day. The previously used petri dishes were saved along with sugarcane leaf pieces and moist cotton ball to determine egg hatch. This ensured similar age for the hatched larvae as well as determination of correct numbers of days for egg hatch. Newly hatched larvae were used either for life cycle studies as mentioned above or preserved in 70% alcohol for morphological descriptions.

For the longevity test, adults were placed individually in petri dishes and provided every other day with 20–30 aphids of mixed ages on small sugarcane leaf pieces. A moist cotton ball also was put in each petri dish as a source of moisture and to delay the desiccation of sugarcane leaf pieces. There were 10 replicates (individual adults) in this experiment, and the experiment was terminated when all adults had died.

Description of *D. terminatus* Larvae. The following measurements were recorded from specimens preserved in alcohol: head length (clypeus to occipital foramen), head width at level of stemmata, maximum body width and length of normally extended specimens. Measurements were made using calibrations on drawing paper superimposed on specimens via a camera lucida mounted on an Olympus SZH10 stereomicroscope at 70× magnification. Larvae of each of the four instars were measured, and results are presented as means and ranges. First and fourth instars also were examined using an Olympus BX50 compound microscope. Fourth instars are described and illustrated in detail. Characters specific to first instars are described and illustrated with special attention to secondary setae. Observations were made at 200–400×, and drawings were prepared using a camera lucida. Habitus illustrations were prepared by drawing lateral halves of specimens as a series of separate drawings. These were inked, scanned, and then reduced and assembled for bilateral symmetry using Adobe Photoshop (Adobe Systems, Mountain View, CA). Bilateral symmetry was achieved by duplicating bilaterally reversed images and splicing them at midlines. Specimens were prepared for microscopic examination by clearing in warm (50°C) 10% KOH aqueous solution, washing in alcohol, and slide mounting in glycerin. Larval terminology follows that of Ślipiński (2007). Voucher specimens are deposited in the Louisiana State Arthropod Museum. Abbreviations used include

T1–T3 (thoracic segments 1–3) and A1–A9 (abdominal segments 1–9).

Assessment of *D. terminatus* as a Biological Control Agent. The potential of larval *D. terminatus* as a biological control agent of *M. sacchari* was assessed by dividing the total number of aphids consumed/killed by the number of days for larval development. Potential of adults was assessed through voracity tests. In this test, individual beetles were starved for at least 24 h, and then each beetle was provided 30 *M. sacchari* nymphs of mixed ages on three or four 5-cm pieces of sugarcane leaves from an aphid-susceptible variety (L 97-128) that was grown in the greenhouse. A small piece of moist cotton ball also was placed inside to avoid desiccation of leaf pieces. There were 15 replicates (individual adults) of this experiment, including three controls with 30 nymphs on pieces of sugarcane leaves added without beetles to assess natural mortality. The numbers of aphids killed in the treatment or dead in the control were recorded after 24 h, and voracity was calculated using the following formula from Soares et al. (2003):

$$V_o = (A - a_{24})ra_{24}$$

where V_o is the calculated number of aphids eaten in 24 h (adjusted for aphid mortality in the controls), A is number of aphid available, a_{24} is number of aphids alive after 24 h, and ra_{24} is the ratio of aphids found alive after 24 h to the initial number in the control treatment.

Data Analysis. Data on size for each developmental stage, and days for egg hatch, larval and pupal development, adult longevity, and total aphids killed by the larvae or adults were subjected to Proc Means (SAS Institute 2005).

Results and Discussion

Life Cycle of *D. terminatus*

The field-collected larvae pupated inside the rearing chamber on pieces of sugarcane leaves rather than on wax paper or Kimwipes. This is contrary to observations by Hall (2001), Hentz and Nuessly (2002), and Hallborg (2003) that wax paper or Kimwipes were the preferred pupation sites. Beetles were provided with one of their natural preys (i.e., *M. sacchari*) on sugarcane leaves in the current study, whereas in previous studies although the aphids were provided, sorghum leaves were not always provided, which might have affected the beetles' choice of a pupation site. *D. terminatus* laid eggs singly, primarily on the sugarcane leaf. Egg deposition on wax paper or Kimwipes was rare, and the few deposited on the bottom of petri dishes failed to hatch. The eggs were usually deposited near the leaf midrib and on the underside of leaf pieces. Length of the convex and elongate eggs was 0.67 ± 0.03 mm (range, 0.58–0.76 mm). Hentz and Nuessly (2002) and Hallborg (2003) reported similar measurements for *D. terminatus* eggs. In the current study, the egg stage lasted an average of 4.5 ± 0.09 d (range, 4.3–4.7 d) (Table 1). Hallborg (2003) reported 6.3 and 6.2 d for the egg stage duration when beetles were fed on *A. gossypii* or *M. persicae*, respectively, and incubated at 22°C (versus 26°C in this study). Hall (2001) observed ≈3 d for egg stage duration at 27.7°C when beetles were fed *S. flava.* The differences in temperature and prey species might have caused these observed variations in egg stage duration. Although sugarcane leaf pieces had desiccated by day 4, eggs were still able to hatch. Fecundity was not recorded in the current study, but Hall (2001) determined that *D. terminatus* laid 3.0 eggs per day for 17.0 d, for a total mean of 42 eggs per female when fed *S. flava.*

Table 1. Number of days of *D. terminatus* at specific stages of development on *M. sacchari* nymphs feeding on sugarcane leaves

Stage (no.)[a]	Days (± SEM)
Eggs (28)	4.50 (0.09)
Larvae	
First instar (21)	1.66 (0.10)
Second instar (18)	1.61 (0.12)
Third instar (18)	1.77 (0.10)
Fourth instar (17)	1.70 (0.18)
Total larval development (24)	6.79 (0.55)
Pupae (19)	4.89 (0.18)
Total larvae to adult (16)	12.12 (0.59)
Adult (10)	26.1 (1.9)

[a] Figures in parentheses indicate the number of individuals as replicates.

The numbers of days for the other developmental stages are given in Table 1. On average, each of the four instars lasted <2 d. The last instar formed a prepupa, most of which were attached to the underside of the sugarcane leaf near the midrib. The larva attached itself to the leaf with a sticky substance released from the abdomen. The last instars sometimes also were seen attaching to the glass wall of vials, but those individuals were unable to pupate. On average, 6.79 ± 0.55 d (range, 5.65–7.93 d) in the larval stage was recorded. However, Hall (2001) reported a 10-d duration at 27.7°C, whereas Hentz and Nuessly (2002) reported 4 d at 27.5°C for the larval stage while feeding on *S. flava.* Hallborg (2003) reported 9.4- and 7.4-d duration in the larval stage for *D. terminatus* when either *A. gossypii* or *M. persicae*, respectively, were used as prey at 22°C. In the current study, the pupal stage lasted an average of 4.89 ± 0.18 d (range, 4.50–5.28 d). Hall (2001) and Hentz and Nuessly (2002) reported similar pupation time (4–5 d) when *S. flava* was used as prey. Hallborg (2003) reported 6.4 and 4.1 d in the pupal stage for *D. terminatus* when either *A. gossypii* or *M. persicae*, respectively, were used as prey. From larval hatch to adult emergence, the current study reports an average of 12.12 ± 0.59 d (range, 10.86–13.38 d) at 26°C. The differences in larval and pupal growth periods in various studies are probably attributable to different prey species, incubation conditions, or both.

In the adult longevity test, an average life span of 26.1 ± 1.9 d (range, 21.9–30.3 d) for *D. terminatus* adults (Table 1) was recorded, but other studies have shown a survival of 143, 75, and 30 d when fed on *A. gossypii, M. persicae*, or *R. maidis*, respectively (Hall-

Table 2. Size and range (in millimetrs) of different stages of *D. terminatus* reared on *M. sacchari* feeding on sugarcane leaves

Stage (no.)	Head length	Head width	Body length	Body width
Eggs (10)	—[a]	—	0.67 (0.58–0.76)	—
Larvae				
First instar (15)	0.14 (0.12–0.15)	0.20 (0.16–0.20)	1.07 (0.70–1.40)	0.37 (0.20–0.50)
Second instar (2)	0.18 (0.16–0.20)	0.29 (0.28–0.30)	2.15 (2.00–2.30)	0.75 (0.70–0.80)
Third instar (2)	0.23	0.39 (0.35–0.42)	2.80	0.80
Fourth instar (3)	0.23 (0.20–0.25)	0.39 (0.38–0.40)	3.00 (2.70–3.50)	1.28 (1.20–1.35)
Pupae (19)	—	—	1.41 (1.25–1.56)	0.76 (0.65–0.87)
Adult (15)	—	—	1.73 (1.59–1.87)	—

If ranges are not given, no variation was evident.
[a] Data not recorded.

borg 2003) and 50 d (Hentz and Nuessly 2002) or 17 d (Hall 2001) when fed on *S. flava.* Hallborg (2003) also reported that adults could survive on as little as one *R. maidis* per day for 10 d. This variation in adult survival may be attributed to different prey; different incubation conditions, such as temperature; or a combination.

Description of *D. terminatus* Larvae

Size measurements of head and body for various life stages are given in Table 2.

First Instar (Fig. 1; Table 2). *Body.* Fusiform, gradually broadened from head to A2–A3 then tapering evenly to A8. Color mottled light gray to brown, with coarse asperites dorsally, fine asperites ventrally. Lateral lobes of body wall less prominent than on fourth instars. Dorsal secondary setae similar in size to homologous setae on fourth instar, so proportionally much larger relative to overall body size. Legs longer relative to body than on fourth instar. Primary setae apparently absent from thoracic nota.

Head. Occiput bearing a pair of large medially curved frayed and serrate secondary setae (possibly egg bursters). Two pairs of frayed, jagged, secondary setae present in postfrontal area, and a single pair of jagged goblet shaped secondary setae present just medial to stemmata.

Thorax. Pronotum with three pairs of blunt, jagged secondary setae in a submedian row. Postmedian area of pronotal disc with a pair of large goblet shaped secondary setae, each borne on a low, sclerotized chalaza. Two pairs of smaller, jagged, goblet setae present, one near middle of disc, the other near anterior lateral margin. Lateral margin with six pairs of jagged secondary setae of varying sizes and shapes.

Mesonotum and metanotum similar with a median raised area bearing a pair of large goblet setae as on prothorax, and a row of four jagged setae along lateral margin of raised area. Lateral margins each with three pairs of jagged setae, the first two approximate, curved and serrate, the third goblet shaped.

Pro-, meso-, and metaventrites each with a single submedian pair of primary setae.

Abdomen. Abdominal segments A1–A8 similar, with four pairs of small fan shaped secondary setae, the median two pairs in a transverse line, the lateral two pair in a longitudinal line. Each lateral lobe with a goblet seta borne on a low tubercle and a jagged curved seta ventral to it. Openings of repugnatorial glands not visible. A9 circular in dorsal view, bearing a postmedian pair of fan-shaped secondary setae, four pairs of jagged setae on lateral and posterior aspect of disc, and three pairs of long primary setae along posterior margin, the longest pair distinctly clubbed apically.

Abdominal ventrites each with three pairs of primary setae in transverse rows, the middle pair shorter than either the median or lateral pair. Each segment with a single primary seta located along lateral margin ventral to lobe.

Fourth instar (Fig. 2; Table 2). *Body.* Fusiform, gradually broadened from head to A2–A3 then tapering evenly to A8, live larvae not covered by waxy exudate. Color of head, mouthparts, legs, and pale brown, dorsal surface of integument brownish gray with darker granulations, imparting a medium gray to gray-brown color overall, lateral lobes of all segments lighter in color than discs. T1 evenly light grayish brown; T2–T3 darker brown, especially in median two thirds; A1–A5 gray-brown with vaguely defined darker brown areas laterally. A6–A8 evenly medium gray-brown. Ventrally light gray. Thoracic nota lacking sclerotized plates. Dorsal integument covered with fine spiny asperites. Dorsal secondary setae of body stout, blunt, ragged along shaft and often with jagged apices, not borne on tubercles or other specialized processes. Distributed evenly or in irregular groups throughout dorsal integument. Secondary setae absent ventrally and from legs and mouthparts. Primary setae normally aciculate on body and mouthparts, tarsugular setae clubbed. Ventral integument with granulate asperites that are much finer than dorsal asperites.

Head. Weakly hypognathus, broader than long, arcuate across anterior face to stemmata, then straight and weakly convergent to occuput. Surface microgranulate, dull. Epicranial stem absent. Three stemmata on each side, arranged in a close triangle. Antennae (Fig. 3a) three-segmented with relative antennomere lengths from base to apex 0.5, 1.0, 0.5. Antennal base broad, membranous. Segment one simple. Segment 2 with three subapical and three apical setae and a conical sensorium that extends 2× length of segment 3. Segment 3 bearing one long seta and three shorter setae. Labrum triangular, anterior mar-

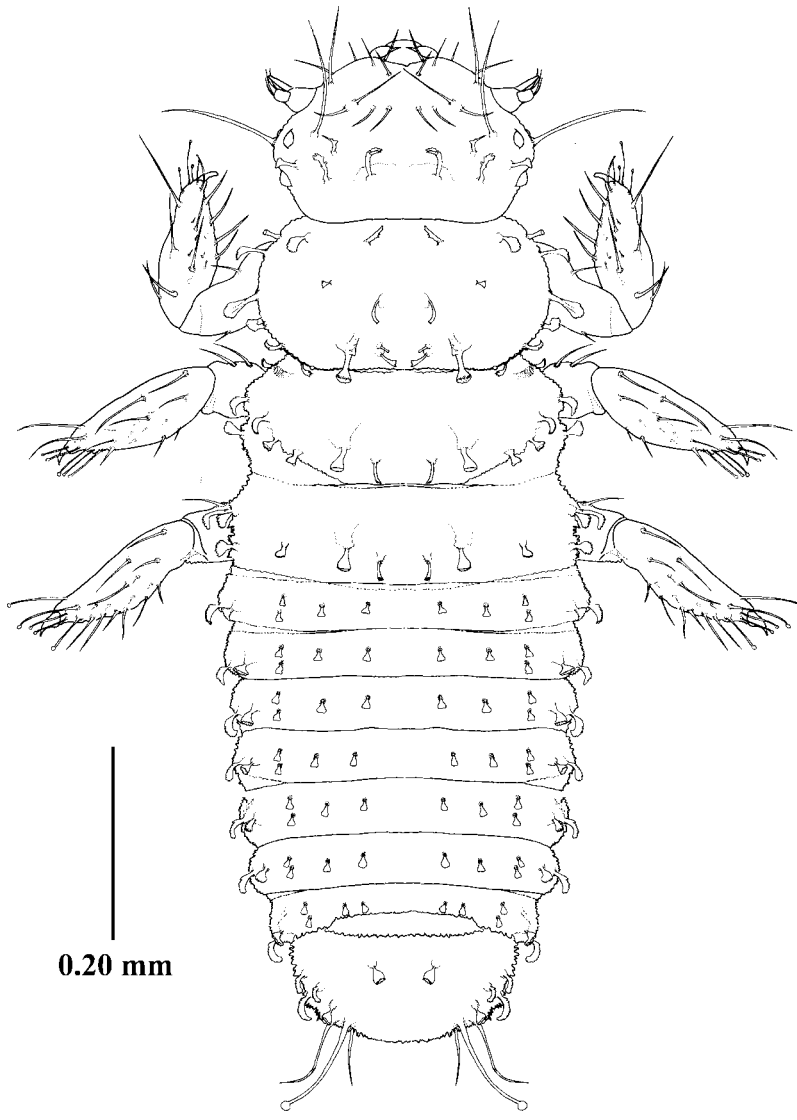

Fig. 1. *D. terminatus*, first instar larva, dorsal habitus. Integumental asperites omitted.

gin straight, posterior margin convergent to angular apex. Mandible (Fig. 3b) simple, apically acute, with shallow incisor groove and flat, straight mola. Scrobe with a single short seta. Hypostomal ridge strong and distinct. Maxilla (Fig. 3c) with rounded, simple mala bearing three sublateral and one distal setae. Maxillary palpi two-segmented, segment 1 broadly triangular, with a single strong seta at apicolateral angle; segment two narrower and 1.5× longer than 1, with a single small seta along medial margin and a clump of sensory papillae apically. Mentum/submentum quadrate, with basal and distal pairs of setae, palpifer distinct. Labial palpi simple, one-segmented with single basal seta and terminal cluster of sensillae.

Thorax. Prothorax with a row of three transverse pairs of primary setae near anterior margin, and four pairs along lateral margin.

Meso- and metathorax similar in length and width, with low, transverse oval elevated area in middle two thirds and two broad lobes laterally on each segment, anterior lobe bearing two primary setae, posterior lobe bearing one seta.

Legs well developed, five segmented, widely separated, each with five to seven clubbed setae arising near apex of tibiotarsus in addition to typical primary setae.

Abdomen. Abdominal segments A1–A8 similar, lacking elevated median areas, lateral lobes single, each bearing a pair of primary and numerous secondary setae. Paired gland openings present along anterior margins of A1–A8. A9 circular in dorsal view, bearing four long primary setae along posterior margin and additional four pairs of shorter setae along margin and deflexed ventral submarginal aspect.

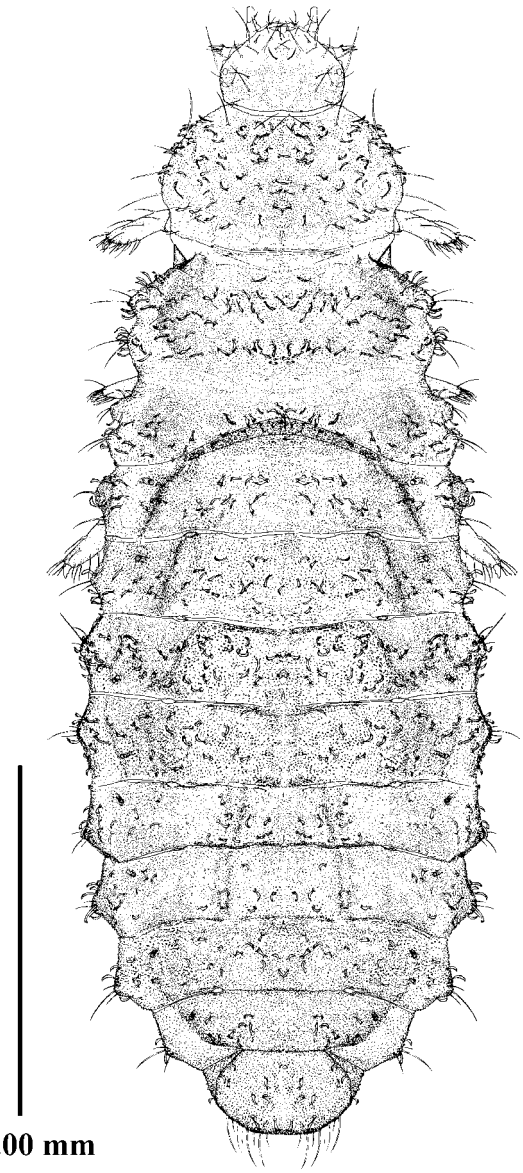

Fig. 2. *D. terminatus*, mature fourth instar larva, dorsal habitus.

Spiracles annular, simple, borne laterally on T2 and dorsolaterally on A1–A8.

Primary setae of ventrites smaller and more slender than dorsal setae, each segment bearing a submedian pair.

Potential of *D. terminatus* as Biological Control Agent

Although 10–15 aphids were provided for each first instar, only an average of 7.71 ± 0.38 aphids (range, 6.17–9.25 aphids) were consumed. The larvae on average consumed a total of 29.88 ± 1.81 aphid nymphs (range, 26.04–33.72 nymphs) for complete development with a consumption rate of 4.65 ± 0.38 aphids per day (range, 3.85–5.45/d). The aphids killed by the larvae were almost always lying upside down and either all of their ventral body parts were consumed or just body fluid was sucked up.

In the adult voracity test, there was no mortality in the control. The adults killed a maximum of 23 aphid nymphs, but the average for 12 beetles was 19.08 ± 0.89 aphid nymphs per d (range, 17.10–21.06). The consumption rates of *D. terminatus* vary when other species were used as prey. Hall (2001) observed *D. terminatus* consuming 5–10 *S. flava* per day, whereas Hallborg (2003) cited average daily consumption rates of 13.5 *A. gossypii* and 8.7 *M. persicae.* But the specific stage (i.e., nymph or adult) of the prey aphid was not mentioned in those reports. The size of the prey also affects the numbers consumed by the coccinellids (Hodek 1996). Only nymphs were used in studying larval development and adult voracity in the current study. A few cursory observations of the feeding behavior of the beetle indicated that one adult beetle took ≈3 min to devour the whole aphid body. Mostly, the adults consumed the whole aphid but sometimes just sucked up the aphid body fluids and left the exoskeleton. A common observation was that beetles moved around randomly for several minutes before attacking the next aphid.

The food consumption rate of coccinellids is affected by several environmental factors, including temperature. Isikber and Copland (2001) reported an increase in the consumption rate of *Scymnus levaillanti* (Mulsant) and *Cycloneda sanguinea* (L.) on *A. gossypii*, with increase in temperature from 25 to 30°C. The current studies were conducted at 26°C, which might have undermined the daily consumption rate of this beetle because temperature generally stays above 30°C during summer days in Louisiana. However, data are not available for comparisons of *D. terminatus* consumption rate at different temperatures or to other coccinellids feeding on *M. sacchari.* Furthermore, the size of the predatory conccinellids affects the number of aphids consumed (Hodek 1996, Isikber and Copland 2001). The extremely small size of larvae as well as adults of *D. terminatus* is a possible explanation for the small number of aphids consumed.

The current commercial varieties of sugarcane in Louisiana sustain very low populations of *M. sacchari.* Greenhouse studies have shown an r_m (intrinsic rate of aphid increase) value as low as 0.05 on the resistant variety HoCP 91-555 or as high as 0.15 on the susceptible variety L 97-128 (W.A., unpublished data). Predation of *D. terminatus* larvae on *M. sacchari* was first noticed in a small plot variety test on 10 July, although aphids were monitored biweekly starting in early April. The abundance of *D. terminatus* seemed to coincide with the peak population time for *M. sacchari* in Louisiana sugarcane, late June through July (McAllister et al. 2005; W.A., unpublished data). With low numbers of aphids and effectiveness of *D. terminatus*, chemical insecticides might not be needed for *M. sacchari* control. However, careful consideration of beneficials such as *D.*

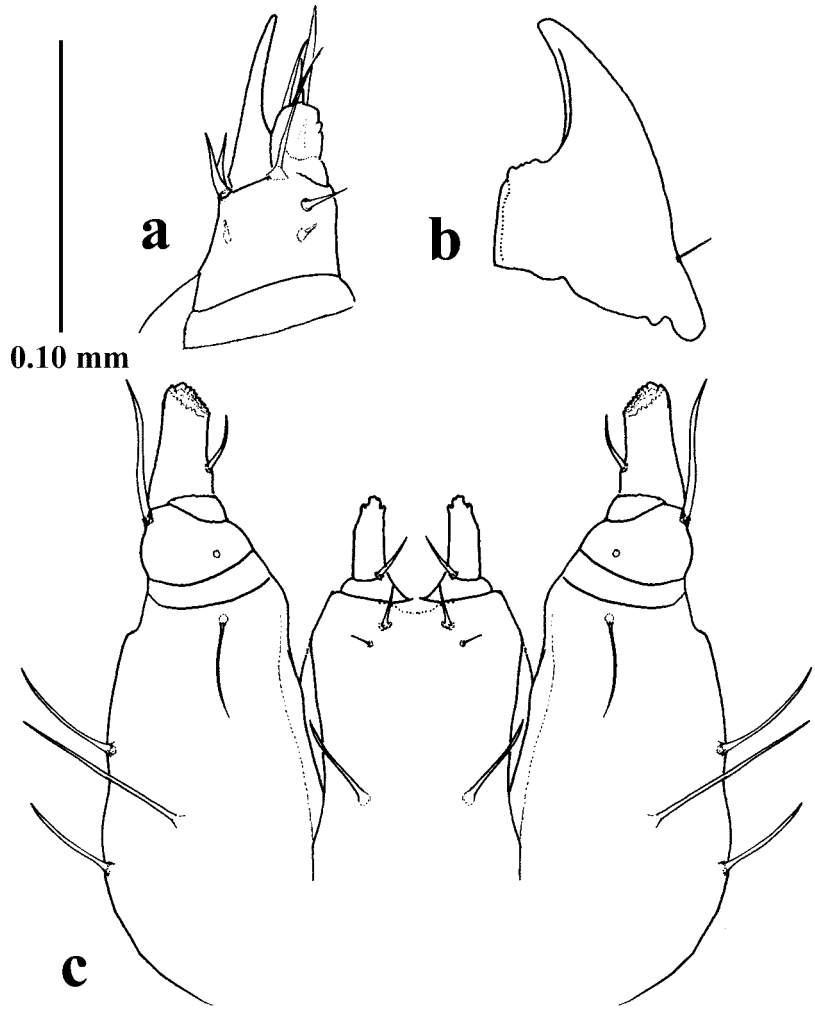

Fig. 3. *D. terminatus*, mature fourth instar larva, details of head. (a) Antenna. (b) Mandible. (c) Ventral mouthparts.

terminatus is important in the development of any new chemistry for managing major insect pest problems in Louisiana sugarcane.

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