PHLEBOTOMINE SAND FLIES OF NORTH AMERICA (DIPTERA: PSYCHODIDAE)†

D. G. YOUNG§ and P. V. PERKINS§

ABSTRACT. Fourteen species of Lutzomyia sand flies are recorded from North America, north of Mexico. Two of them, L. apache n.s.p. and L. taquapitica n.s.p. from Arizona, are described for the first time. Lutzomyia diabolica is treated as a valid species, distinct from L. cruciata. Lutzomyia vexator, having a widespread geographic distribution in North America, is considered as a monotypic taxon without subspecies. Identification keys, distributional data, references, biological and disease information are provided.

INTRODUCTION

During the past decade, interest in North American sand flies has increased due to confirmed reports of autochthonous human leishmaniasis in Texas (Shaw et al. 1976), canine leishmaniasis in Oklahoma (Anderson et al. 1986), and Rio Grande virus in Neston woodrats in Texas (Callisher et al. 1977). Phlebotomines are the suspected, but as yet unproven, vectors of these diseases in these foci.

This report summarizes the published and unpublished information on the taxonomy and geographic distribution of the known species of Lutzomyia in North America (north of Mexico), including information on their bionomics and vector potential where known. References and identification keys are provided.

Coquillet (1907) first reported sand flies in the Western Hemisphere. He described Lutzomyia cruciata (as Flebotomus cruciatus) from Guatemala and L. vexator (as Flebotomus vexator) from Plummer's Island, Maryland. Shannon (1913) observed L. vexator females feeding on snakes at the type-locality and speculated that another female had bitten a person sleeping inside of a cabin.

A group of biologists from Cornell University were tormented at night by biting sand flies on Minnie's Island, Okfuskee County, Oklahoma in 1912. Johannsen (1943) recalled that a "member of the party counted over 75 punctures on his body, another's hands and arms were swollen to abnormal size..." Local residents, acutely aware of these insects, called them "merye wings," an Elizabethan English term reflecting the origin of most of the residents. Johannsen (1943) studied one defective sand fly (lot no. 1981), concluding that it differed from L. vexator in wing venation and palpal proportions but he did not further describe the specimen which has since been lost (Quentin Wheeler, in litt., 1982). The identity of these man-biting sand flies remains unknown, but they probably represented L. shannoni (Dyar) and/or L. cruciata.

Apart from this report and from man-biting observations of L. shannoni in the southeastern USA, the only other area in North America where sand flies commonly bite people is in southcentral Texas. Parman (1919) observed a species, later named Phlebotomus diabolus Hall 1936, biting people at Uvalde, Texas. Lindquist (1936) added more information on the habits of this species and reported that females attacked man at night in dwellings from 2000 to 2400 hr. We now know that L. diabolus will attack man in the daytime as well.

PHLEBOTOMINE-BORNE DISEASES IN THE UNITED STATES

Cutaneous leishmaniasis in humans

Simpson et al. (1968) documented the first parasitologically-confirmed human case of autochthonous leishmaniasis in the USA. Leishmanias were observed in tissue smears and culture medium. The patient, who suffered from diffuse cutaneous leishmaniasis, had lived in San Benito, Cameron Co., Texas for her entire life except for occasional visits to the Mexican border states of Tamaulipas and Nuevo León. Subsequently, Shaw et al. (1976) discussed 2 additional human cases of cutaneous leishmaniasis in Texas (Gonzales and Dilworth counties). They believed that the disease had been locally acquired in both cases. Walton et al. (1977) identified one of the Texas strains (no. 1156) as belonging in the Leishmania mexi-
cana complex. Peters (1981), referring to the Texas Leishmania as Leish. mexicana pifanoi, suggested that the disease may have been introduced from Venezuela. Kreutzer et al. (1983) identified one Texas strain (WR 127) as Leishmania mexicana mexicana. Parasites were cultured from a boy with a facial lesion, who probably contracted the disease in the vicinity of Uvalde, Texas in 1981 (Larry Hendricks, personal communication). Three unrelated persons from or near San Antonio, Texas, contracted cutaneous leishmaniasis in 1982 and it is believed the infections were also locally acquired (Tracy Gustafson, personal communication). In view of these reports, there is a good possibility that Stewart and Pichner (1945) earlier had correctly diagnosed cutaneous leishmaniasis in a boy who lived near Alice, Texas.

The epidemiology of leishmaniasis in Texas, though not yet studied, may be similar to that in nearby Coahuila State, Mexico where Ramos-Aguirre (1970) discussed 3 autochthonous human cases, 2 of which were of the diffuse form, similar to that reported by Simpson et al. (1968). Díaz Nájera (1971) collected specimens of L. diabolica at Múzquiz, Coahuila, a site where one of the patients lived. This sand fly, the only species known to bite people in Texas and northern Mexico, is the suspected vector. The nonforested, relatively dry habitat in these areas differs greatly from classical Leishmania mexicana foci in tropical forests of southern Mexico, Central and South America.

Visceral leishmaniasis in dogs

MacVean et al. (1979) and Anderson et al. (1980) reported visceral leishmaniasis in hunting dogs (fox hounds) from a kennel at Edmond near Oklahoma City, Oklahoma. The disease continues to be transmitted to dogs at this site, but its origin remains unknown. The causative agent is similar to Leishmania donovani infantum, a parasite causing canine and human visceral leishmaniasis in the Old World (Decker-Jackson and Tang 1982, Kocan et al. 1983), but Kreutzer et al. (1985) identified the parasite as Leishmania mexicana. MacVean et al. (1979) captured specimens of Lutzomyia near the kennel in the forest. We provisionally identified one of the males as L. vexator, on the basis of a drawing of the gonostylus kindly given to us by R. A. Ward, Walter Reed Army Institute of Research. The vector of leishmaniasis at this site remains unknown. Leishmania-infected dogs are occasionally imported into the United States and Canada from endemic areas (Maness 1981, Simpson et al. 1982) and there is recent serological evidence that coyotes from Texas may be infected with the disease (Grogi et al. 1984).

Rio Grande Virus in vertebrates

Rio Grande virus, family Bunyaviridae, genus Phlebovirus (Bishop et al. 1980), was named by Calisher et al. (1977) who recovered the virus from Neotoma woodrats near Brownsville, Texas and found serological evidence of infection in other vertebrates but not in people. Lutzomyia anthophora (Addis) is the suspected vector based on the habits of the species, its close association with woodrats, and its proven ability to vertically transmit the virus in the laboratory (Endris et al. 1983).

Parasites of amphibians and reptiles

Ayala (1973) gave references to his earlier, detailed studies on amphibian and reptile parasites associated with phlebotomines, especially L. vexator, in central California. He showed that sand flies were suitable hosts for Trypanosoma bufophlebotomi Ayala of toads and T. scelopori Ayala and T. gerronti Ayala and McKay of lizards. A saurian malaria, Plasmodium mexicanum Thompson and Huff, developed to the sporozoite stage in sand flies and was infective when inoculated into Sceloporus fence lizards. Wild-caught sand flies also were found infected with hemogregarines (Hepatozoon sp. oocysts) that infected both lizards and snakes following experimental inoculations (Ayala 1970a).

METHODS AND MATERIALS

FIELD COLLECTIONS

The diversity of the sand fly fauna in a given area is best determined by using a combination of collection techniques. Such techniques were discussed by Lewis (1973, 1974) and Chaniotis (1978). In the present study, adult sand flies were routinely searched for in their diurnal resting sites, including tree trunks, under loose bark of dead standing trees, tree hollows, animal burrows and dens, rock crevices and other protected microhabitats. Specimens were captured with a simple tube aspirator, burrow traps and sticky paper traps. Endris et al. (1982) outlined procedures for handling such wild-caught sand flies for rearing purposes.

Flight traps and CDC light traps were used whenever possible (see Gessitt and Gessitt 1962, Sudia and Chamberlain 1962). They are effective devices for capturing most, but not all species. Limited attempts to recover immature
stages from soil samples using a Berlese funnel were unsuccessful. As far as is known, immatures have not been found in nature in North America.

**Preservation and Slide Mounting**

It is recommended that adult sand flies be stored dry between layers of tissue paper in a pill box before they are mounted on slides because specimens preserved in alcohol are much more difficult to macerate, particularly those that have been stored in the preservative for a year or more. Following maceration in 10% NaOH, however, the flies can be stored indefinitely in 70% EOH. Methods for preparing slide mounts and identifying sand flies are outlined below following Young (1979).

1. Place whole sand fly in 10–25% sodium hydroxide (NaOH) at room temperature in a small crucible for 30–60 minutes.
2. Heat the NaOH to the boiling point on a hot plate, then immediately remove crucible from heat and allow to cool for 30 minutes.
3. Put macerated fly directly into a drop of 90% liquid phenol in the depression in a depression microscope slide (temporary slide mount) and identify with the aid of a compound microscope. A coverslip can be placed over the specimen if necessary. The internal spermathecae of female sand flies should then be clearly visible.
4. Preserve in 70% alcohol in a vial, or proceed to step 5 if a permanent slide mount is needed.
5. Place one drop of Canada balsam in the depression of another microscope slide. Mix 2–3 drops of phenol with it and put specimen or specimens (up to 12) in the mixture.
6. Allow the phenol to slowly evaporate at room temperature until the balsam-phenol mixture becomes somewhat viscous. The mixture has now infiltrated the body of the sand fly and the spermathecae and other structures should be preserved.
7. Dissect the specimen by removing the head and wings. Lift these parts and the remainder of the specimen with a small needle and position them on a coverslip in small drops of the balsam-phenol mixture in which the fly was disinfected. Orient the head so that the cibarium can be viewed as in the illustrations in this paper.
8. Place a small piece of glass, from a previously ground coverslip, in each corner of the coverslip holding the dissected sand fly. Allow to dry until the drops of mixture are completely hard.
9. Invert coverslip over a drop of Canada balsam placed in the middle of a clean microscope slide. Store the slide horizontally until dry.

**Material Examined**

A number of colleagues generously supplied specimens, locality records (specimens not examined by the present authors), and other pertinent information. We gratefully acknowledge the help of F. S. Blanton, J. F. Butler, R. G. Endris, G. B. Fairchild, R. C. Johnson, W. L. Kramer and R. C. Wilkerson, all associated with the University of Florida at the time of this study; J. F. Reinert, R. W. Intermill, P. G. Law- yer and J. I. Glick, US Army Medical entomologists; R. H. Roberts, ARS, USDA, Gainesville; W. W. Wirth, US Department of Agriculture; R. J. Brenner and S. Frommer, University of California, Riverside; Q. D. Wheeler, Cornell University; L. D. Hendricks and R. A. Ward, Walter Reed Army Institute of Research; Personnel of the US Army Health Services Command at various posts in the continental USA and C. G. Moore and D. B. Francy, Centers for Disease Control, Ft. Collins, Colorado.

Dr. V. F. Newhouse, Centers for Disease Control, deserves special recognition for allowing us to publish his records of North American sand flies identified earlier by G. B. Fairchild and W. J. Hanson but not confirmed by the present authors.

Holotypes of all new species will be deposited in the US National Museum of Natural History along with paratypes and other specimens. Paratypes also will be deposited in the collection at the University of Florida, and elsewhere as indicated in the text.

**Taxonomic Treatment**

Many of the species of *Lutzomyia* were originally described in the genus *Plebotomus* Ron- dani and Berté (= *Flebotomus*), a name now applied strictly to the medically-important group of Old World phlebotomines (Theodor 1965). The subgeneric and species group classification of *Lutzomyia* follows that of Lewis et al. (1977) and Martins et al. (1978). The morphological terminology agrees with that used by Quate and Vockeroth (1981). Thus, gonostylus and gonocoite are substituted for style and coxite, respectively. The ejaculatory apodeme and sperm pump together refer to the familiar term, genital pump, that is used by many students of Phlebotominae. For quick reference, the structures shown in Fig. 2 are labelled. The immature stages are not treated in this review because of a lack of material, but references to them are given. Well known species are not redescribed because adequate descriptions are available in other papers cited here in each species account.

**Systematic Account**

**Check list of North American Phlebotominae**

**Genus *Lutzomyia*** França 1924

**Subgenus *Lutzomyia*** França 1924
1. *L. cruciata* (Coquillett 1907)
2. *L. diabolica* (Hull 1930)
Subgenus *Ceromyia* Barretto 1962 (= Species Group *vesperilomia* Theodor 1963)
3. *L. aquatilis* (Fairchild and Harwood 1961)
Subgenus *Dempfoomyia* Addis 1945
4. *L. anaxiophora* (Addis 1945)
Subgenus *Pseudomyia* Barretto 1962 (= Species Group *shannoni* Theodor 1965)
5. *L. spheniolepis* (Dyar 1929)
6. *L. tanyophillus* Young and Perkins n.sp.
Species Group *aragani* Theodor 1965
7. *L. texana* (Dampf 1898)
Subgenus *Helocyrtomyia* Barretto 1962 (= Species Group *vexator* Theodor 1965)
8. *L. apache* Young and Perkins n.sp.
9. *L. opulenta* (Dampf 1944)
10. *L. stewarti* (Mangabeira and Galindo 1944)
12. *L. vexator* (Coquillett 1907)
13. *L. california* (Fairchild and Hertig 1957)
14. *L. subensis* (Fairchild and Trappido 1950)

KEY TO THE NORTH AMERICAN SPECIES OF *LUTZOMYIA*

It is usually necessary to prepare slide mounts of sand flies in order to study the structures of taxonomic importance. Once the local fauna becomes known, however, it is often possible to identify specimens simply by using color characters, relative lengths of the legs, body, wings and other features which can be seen without the aid of a microscope. In general appearance, most sand flies resemble the one shown in Fig. 1. Sexual dimorphism is marked. The male has conspicuous external terminalia, its abdomen is relatively slender and it is usually smaller than the conspecific female.

**Males**

1. Gonostylus of genitalia with 5 large spines; subterminal seta absent. Gonocoxite with a basal tuft or group of persistent setae

2. Gonostylus with 2–4 large spines; subterminal seta and gonocoxal tuft present or absent

3. Cibarium with a row of 8 or more short horizontal teeth. Gonocoxal tuft of 10–15 setae. *L. californica* (Fig. 14)

4. Paramere with dorsal setae restricted to apical third. Aedeagal filaments beyond apical inflated portion shorter than inflated portion. *L. oppidana* (Fig. 11)

5. Lateral lobe barely extending beyond end of paramere. Gonostylus with paired basal spines inserted at same level; subterminal spine close to terminal pair. Antennal ascodis on flagellomere II shorter than one-third the length of flagellomere. *L. stewarti* (Fig. 12)

6. Antennal ascodis on flagellomere II longer than half the length of flagellomere

7. Antennal ascodis on flagellomere II extending beyond end of flagellomere

8. Gonostylus with 2 or 3 strong spines; subterminal seta present. Paramere with acute ventral projection

9. Gonostylus with 4 strong spines; subterminal seta absent. Paramere with or without acute ventral projection


11. Paramere simple, without a dorsal arm. Gonostylus with proximal spine about the same size as others. Gonocoxite with basal tufts of setae. Lateral lobe inflated. Aedeagal filaments longer than 3 times length of spermatheca

12. Antennal ascodis with distinct proximal spurs. Palpomere 5 shorter (Figs. 8, 9)

13. Antennal ascodis simple, without proximal spurs. Palpomere 5 longer (Figs. 4, 12)

14. Gonocoxite with numerous setae at middle and beyond. Lateral lobe longer than coxite. Antennal ascodis extending beyond ends of flagello-
Fig. 1. A female phlebotomine sand fly.
meres, proximal spurs relatively short as shown
Gonoxocite without persistent setae.
Lateral lobe shorter than gonoxocite.
Antennal axes on reaching ends of flagellomeres, proximal spurs much longer ............... L. shannonii (Fig. 9)
10. Gonoxocite lacking persistent setae
...L. cubensis
Gonoxocite with 1 or more persistent setae

11. Aedeagal filaments shorter than twice length of ejaculatory apodeme and sperm pump; filament tips enlarged, each with 5 inner teeth. Gonoxocite with 1–2 setae at inner base. Whole insect pale .............. L. xerophilus
Aedeagal filaments over twice as long as ejaculatory apodeme and sperm pump; filament tips simple, without teeth. Gonoxocite tuft of 4 or more setae inserted on a central base. Scutum dark

12. Pleuron much paler than scutum.
Flagellomere 1 extending to level of palpomere 3. Gonoxocite with 12–16 relatively thick persistent setae. Wings grayish ............... L. cruciata (Fig. 2a)
Pleuron as dark as scutum. Flagellomere 1 relatively short, extending to level of palpomere 2. Gonoxocite with fewer than 10 slender persistent setae.
Wings yellowish ........ L. diabolica (Fig. 3)

Females
1. Pharynx armed with posterior spines .............. L. cubensis (Fig. 15)
Pharynx unarmed
2. Antennal axes with proximal spurs
Antennal axes simple, without proximal spurs
3. Cibarium with 8 or more horizontal teeth. Antennal axes with short proximal spurs as shown in Fig. 9. Spermathecae large and spherical without annulations ............ L. texana
Cibarium with 4 horizontal teeth. Antennal axes with long proximal spur ending near base of flagellomeres. Spermathecae tubular or spherical with basal annulations
4. Spermathecae tubular, much longer than wide, without annulations; common sperm duct longer than individual duct. Head width and height subequal; eyes relatively large as shown in Fig. 7
Spermathecae spherical, as long as wide, with basal annulations; common sperm duct shorter than individual duct. Head much longer than wide; eyes relatively small .......... L. tanypus (Fig. 7)
5. Cibarium with a comb-like row of 14 or more horizontal teeth. ...L. californica (Fig. 14)
Cibarium with 2–10 horizontal teeth
6. Cibarium with 2 broadly rounded or blade-like horizontal teeth and with numerous lateral teeth
Cibarium with 4–10 pointed horizontal teeth; lateral teeth inconspicuous or absent
7. Cibarium with 2 rounded horizontal teeth, each with 1 or 2 short spines. Spermathecae wrinkled, oval without bubble-like evaginations ...L. angolonta (Fig. 5)
Cibarium with 2 blade-like horizontal teeth. Spermathecae modified with numerous bubble-like evaginations ............ L. astrophora (Fig. 6)
8. Individual sperm ducts shorter than stem of genital fork. Spermathecae with an outer envelope arising from base of spermathecae .....L. xerophilus (Fig. 4)
Individual sperm ducts longer than stem of genital fork. Spermathecae differently shaped and lacking an outer envelope
9. Spermathecae spherical, spermathecal width the same as, or greater than, width of common sperm duct; with distinct annulations at base
Spermathecae button-like, width less than width of common sperm duct, without basal annulations, but transverse striations or excrescences may be present on individual duct
10. Labrum shorter than combined length of flagellomeres II + III and shorter than twice the length of palpomere 5. Cibarium with only 4–6 very small vertical teeth ............. L. stewarti (Fig. 12)
Labrum subequal to, or longer than, combined lengths of flagellomeres II + III and longer than twice the length of palpomere 5. Cibarium with 10 or more vertical teeth
11. Tergite nine with 2 heavily sclerotized, anterolateral papillate lobes. Labrum shorter than flagellomere 1. Cibarium with small vertical teeth in 2–3 irregular rows; pigment patch shaped as shown in Fig. 2. Individual sperm ducts at least 6 times length of spermatheca. Scutum dark, pleuron pale. Wings grayish .......... L. cruciata (Fig. 2a)
Tergite nine without papillate lobes. Labrum longer than flagellomere 1. Cibarium with 4–7 large vertical teeth in a single row; pigment patch otherwise, relatively broad and darker at expanded base. Individual sperm ducts about 4 times length of spermatheca. Scutum and pleuron pale. Wings yellowish ........ L. diabolica (Fig. 3)

* Applies to specimens from the USA but is not always valid for Mexican and Central American specimens.
12. Individual sperm ducts relatively short, not over 3 times length of stem of genital fork .................. L. weaver (Fig. 8F & 13)

Individual sperm ducts over 5 times length of stem of genital fork ...... 13

13. Antennal scoids of flagellomere II extending beyond end of flagellomere. Individual sperm ducts relatively slender as shown in Fig. 10 ... L. apache (Fig. 10)

Antennal scoids of flagellomere II not extending to end of flagellomere. Individual sperm ducts wider, as shown in Fig. 11 ..................................... L. oppidana (Fig. 11)

SCHWENUS LUTZOMYIA FRANGA

1. Lutzomyia (Lutzomyia) cruciata Coquillett, Fig. 2.

Flebotomus cruciatus Coquillett 1907:102 (♀, Trece Aguas, Alta Vera Paz, Guatemala).


Brumptomyia cruciata: Lewis 1965:575 (internal structures, Belize)


KNOWN DISTRIBUTION: Panama to SE Mexico and USA. Florida (Alachua and St. Johns counties); Georgia (Chariton Co.), Fig. 18.


DISCUSSION: Both sexes of *L. cruciata* from North America have relatively small eyes (Fig. 2) when compared to those of *L. cruciata* from tropical South America. The character state and the slightly thicker setae of the coxite tuft of Florida males support the idea that the neotropical population of *L. cruciata* is an isolated one that may have been derived from neotropical stock that spread through the Gulf of Mexico coastal plain into Florida and Georgia. It is also possible that individuals entered Florida by crossing the Gulf of Mexico, possibly with the aid of hurricane winds, but this hypothesis seems less likely. *Luotzymia cruciata* is not known to occur in the West Indies.

Existing populations of *L. cruciata* in Florida and Georgia are geographically isolated from those in southeastern Mexico, Central America, and northern Mexico. *Luotzymia cruciata* and *L. diabolica* have allopatric distributions in the USA (Fig. 18) but the extent of their geographic distributions in Mexico has not been determined. *Luotzymia cruciata* occurs mainly in secondary and primary hardwood forests, but *L. diabolica* has been found in dry, sometimes treeless localities (e.g., tejas). All of the *L. cruciata* females from Florida and Georgia have a papillate lobe on each side of the 9th abdominal tergite (Fig. 2D). This modification is absent in females of *L. diabolica*, *L. gomlesi* (Nitz.), and some *L. cruciata* females from Mexico, Guatemala, Honduras, and Nicaragua.

In Belize, Lewis (1965) observed that 96% of *L. cruciata* females dissected were parous, thus indicating that the species is normally autogenous. Perkins (1982) established a laboratory colony of *L. cruciata* from a gravid female collected near Gainesville (Alachua Co.), Florida. All females from the F₁ through F₆ generations were autogenous and deposited 35.8 ± 13.5 eggs (range 1–60), 3–10 days following emergence. Other rearing data based on this colony are given by Perkins (1982).

Eighteen females of *L. cruciata* were collected in CDC light traps near Gainesville (Sugarloaf Hammock) in August and September, 1980 (8 trap nights). Five of them, held in a 120 ml feeding chamber (Endris et al. 1982), took blood meals on the arm of a human volunteer during a 1–3 minute period. Their bites were quite painful when compared to those of *L. shannoni*. All 18 females died without depositing eggs and most lived 2.9 ± 2.4 days after capture. The specimens that took human blood meals survived from 6 to 8 days but none had mature eggs when dissected. Lab-reared females were given the opportunity to feed on man on numerous occasions but none, including those that had oviposited 1–7 days previously, did so.

There is little evidence indicating that *L. cruciata* is a natural vector of cutaneous leishmaniasis (*Leishmania mexicana*) in Belize (Williams 1970). Williams (1966), however, demonstrated that *Leishmania mexicana* multiples and develops in *L. cruciata* females and that experimentally-infected flies transmitted the parasite by bite to a human volunteer.

2. *Luotzymia (Luotzymia) diabolica* (Hall), Fig. 3.

*Phlebotomus* sp.: Parmar 1919:211 (biying, man, Texas)


*Lutzymia cruciata diabolica*: Lewis 1975:509 (mouthpart morphol.,). KNOWN DISTRIBUTION: SW Mexico to USA Texas (Atacosa, Bexar, Cameron, Comal, Edwards, Gillespie, Hale, Kinney, Llano, Medina,
Nueces, Sutton, Tom Green, Travis, Uvalde and Val Verde counties), Fig. 18.


Additional records: Texas—2 ♂ (Tom Green Co.), San Angelo Reservoir, 11-VI-1968, light trap, V. Newhouse.

Discussion: Addis (1945b) redescribed L. diabolica from toptotypic specimens, noting that the males had 6 horizontal teeth in the cibarium. Fairchild and Hay (1945a, 1945b) distinguished the male of this species from that of L. cruciata by its more slender ejaculatory apodeme with its cup-shaped proximal end and by the presence of fewer and more slender setae of the gonocoxal tuft. Differences between the females of L. diabolica and L. cruciata were mainly those related to the size and number of vertical teeth in the cibaria, the forms having only 8–10 heavy blunt teeth; the latter with more numerous, smaller vertical teeth. Dorsolateral, papillate lobes of the ninth abdominal tergite were observed in females of both species but were less developed in L. diabolica. Only 4 females of L. diabolica were available to these authors for study.

Disney (1968) studied structural variation in numerous males and females of L. cruciata from Belize, concluding that it was not possible to separate this species from L. diabolica based on available criteria. He observed that the proximal end of the ejaculatory apodeme of was of the "cruciata form" in 80% of the Belize males examined. The basal gonocoxal setae varied in number from 8 to 23 (K = 16), and the number of vertical teeth in the female cibarium ranged from 8 to 29 (K = 16). Other features, including the spermathecae, chitinous arch of the cibarium and number of episternal setae, were studied. From his analysis, he concluded that variation among his material was infraspecific and that the name diabolica should be "pressed, at least as a specific epithet."

In the USA and Mexico, however, there are 2 distinct forms that can be identified as L. diabolica and L. cruciata and which are easily distinguished by the characters given in the keys. Disney (1968) was correct in assigning his Belize specimens to the latter species; L. diabolica is not known to occur there or in other countries south of Mexico but there is a aggressive mantibes that populations exist in western Guatemala.

Luttsomyia diabolica is very dark; the pleuron of each sex is as dark as the scutum, unlike L. cruciata which has a pale pleuron. There seems to be no single, consistent structural difference between the 2 species although the curious papillate lobes of the 9th tergite are present in all L. cruciata females from the USA and in 83% of 81) of L. cruciata females from Mexico and Central America. Females of L. diabolica lack such lobes.

The ratio of the length of flagellomere 1 to the head height (flag. l/head height) of L. diabolica females ranges from 0.45 to 0.61 (n = 31, \( \bar{x} = 0.53, SD = 0.032 \)); that for males ranges from 0.52 to 0.63 (n = 5, \( \bar{x} = 0.568, SD = 0.041 \)). In females of L. cruciata females from Mexico and Central America (n = 73), this ratio varies from 0.61 to 0.88 (\( \bar{x} = 0.792, SD = 0.056 \)); that for L. cruciata males ranges from 0.66 to 0.86 (n = 25, \( \bar{x} = 0.756, SD = 0.048 \)). The number of horizontal teeth in the female cibarium of both species ranges from 4 to 7. Females of L. diabolica are aggressive mantibes, attacking mostly at night but occasionally in the daytime as well. The natural resting and breeding sites of this species are unknown. At Garner State Park, Uvalde Co., Texas, about 1500 specimens were collected from late May to early June 1983 on the inside walls of latrines. The new record of this species from Plainview, Texas, represents the northernmost limit of its known geographic range but few attempts have been made to collect this species in northern Texas, New Mexico or Oklahoma.

Lindquist (1936) and Endris et al. (1982) reared L. diabolica in the laboratory; the former author providing information on its life cycle and describing the immature stages. Females are anautogenous. Additional field studies of this species are recommended in view of its man-biting habits and possible role as a vector.
of cutaneous leishmaniasis in Texas and northern Mexico.

**Species Group Miconia Theodor**

3. *Lutzomyia xerophila* Young, Brenner and Wargo, Fig. 4.

*Lutzomyia xerophila* Young, Brenner and Wargo 1983:313 (♂️, ♀, Cahuilla Hills, Palm Desert, Riverside Co., California).

**Known distribution:** USA, California (Riverside Co.), Fig. 19.


**Discussion:** *Lutzomyia xerophila* is provisionally placed in the *miconia* Group of *Lutzomyia* (Theodor 1965) on the basis of male and female structures, but the male lacks a subterminal seta on the gonostylus unlike other males in the group. This pale species is easily recognized by the characteristic spermathecae of the female and by the male terminalia. Intraspecific variation of the cibarial armature of females is shown in Fig. 4.

Specimens have been taken only in CO₂ traps in the Colorado Desert of Riverside Co., California from May to November. Nothing is known about the feeding habits, diurnal resting sites or biology of this recently described species.

**Subgenus Coromyia Barretto**

4. *Lutzomyia* (Coromyia) *aquilina* (Fairchild and Harwood), Fig. 5.


**Known distribution:** Canada, Alberta and British Columbia, USA, Colorado (Larimer Co.); Washington (Adams and Whitman counties), Fig. 19.


**Discussion:** The small median seta shown on the male gonostylus (Fig. 5A) is absent in the other males of this species examined. The female sperm ducts are illustrated for the first time following immersion of the slide-mounted specimen in 90% liquid phenol for one week.

Shemanchuk et al. (1978) collected 106 specimens of *L. aquilina* in the burrows of the marmot, *Marmota flaviventris nasophora* (Hovore), during a 3-year study in Southern Alberta. Flies were collected from August to October at one site, and in July and August at the Columbia Wildlife Refuge, Washington State (Fairchild and Harwood 1961). The specimens from Colorado were collected near nests of *Neotoma* woodrats located in rock crevices.

The feeding habits of this species remain unknown.

**Subgenus Dampfomyia Addis**

5. *Lutzomyia* (Dampfomyia) *anthophora* (Addis), Fig. 6.


**Known distribution:** Canada, Alberta and British Columbia, USA, Colorado (Larimer Co.); Washington (Adams and Whitman counties), Fig. 19.


**Discussion:** The small median seta shown on the male gonostylus (Fig. 5A) is absent in the other males of this species examined. The female sperm ducts are illustrated for the first time following immersion of the slide-mounted specimen in 90% liquid phenol for one week.

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The feeding habits of this species remain unknown.

Known distribution: Mexico to USA. Texas (Bexar, Cameron, Kinney, Medina, Presidio, San Patricio, Uvalde and Val Verde counties). Fig. 19.


Discussion. In addition to the material noted above hundreds of L. anthophora collected in Neotoma woodrat dens near Brownsville, Texas in 1966, 1975 and 1980 were examined.

The spermathecae with the unusual bubble-like evaginations (Fig. 6l) and the distinctive male genitalia (Fig. 6A) readily distinguish L. anthophora from other species in North America. It is currently impossible to separate females of this species from those of L. dodegi (Vargas and Díaz Nájera) from SW Mexico where the former author (Vargas and Díaz Nájera 1953a) distinguished these females by differences in the cibarial armatures and spermathecae, characters that were found in this study to be unreliable for species identification.

Fairchild and Hettig (1973b) believed that the female of L. dodegi described by Vargas and Díaz Nájera (1953a) actually was a female of L. anthophora. This conclusion was erroneously based on their study of specimens that were later named L. attenuata by De León (1971), not L. dodegi (Porter and Young, unpublished observations). Thus, L. anthophora and L. dodegi will key out together in couplet 2 of Fairchild and Hettig's 1956 key to the species of Dambomyia.

Addis (1945b) reported L. anthophora females feeding on domestic rabbits at Uvalde, Texas. No specimens were taken on other hosts or from suspected resting sites. Young (1972) found both sexes of this species in the dens of woodrats, Neotoma microtus Baird, and concluded that adults were present throughout the year in South Texas. Subsequently, one of us (D.G.Y.) collected more than 600 specimens of L. anthophora from a single woodrat den, east of Brownsville, Texas, on 31 May 1975. It was estimated that an equal number escaped capture. At that time, and in 1980, other specimens were also discovered resting in or near small rodent nests located underneath discarded lumber and cardboard. From these field associations and on laboratory observations, it is suspected that L. anthophora females feed primarily on small mammals; suitable laboratory hosts include hamster (Addis 1945b), white mouse, squirrel (Sciurus), calf, rabbit, opossum (Didelphis) and domestic pig (Endris et al. 1982). Females feed to repletion and are gonotrophically concordant; 33 females (14%) out of 228 females that survived first oviposition in the laboratory took a second blood meal. Several fed a third time and one took a partial fourth bloodmeal before dying (Endris and Young, unpublished observations).

Addis (1945c) and Endris (1982) studied the life cycle of L. anthophora in the laboratory. The former author used a rabbit blood-garden soil mixture for larval food; the latter used a composted rabbit feces-laboratory chow mixture (Young et al. 1981). At 28°C, the mean time from egg deposition to adult emergence was 45 days (n = 72, Addis 1945c) but was only 39 days in the later study (n = 41) (Endris 1982). These conflicting results apparently were due to differences in the larval diet. Addis (1945c) described the immature stages from reared material.

Endris et al. (1983) experimentally infected L. anthophora females with Rio Grande virus that was subsequently recovered from 54.5% of their F1 progeny. This was the first virologically confirmed demonstration of vertical transmission of a Phlebovirus by sand flies. Leishmania mexicana (VARIP strain 411 from Texas) was successfully transmitted to non-infected Syrian hamsters by the bites of experimentally infected, laboratory-reared L. anthophora (Endris and Young, unpublished observations).

Subgenus Psathyromyia Barretto
(= Species Group shannoni)

6. Lutzomyia (Psathyromyia) shannoni (Dyar), Fig. 7.


ADDITIONAL RECORDS (all from D. V. Newhouse, in light traps). Arkansas—1 0 (Union Co.). El Dorado, 21-V-1981. Florida—3 0 (Collier Co.), Corkscrew Swamp, 11-12-IX-1965. 1 0 same data but 9-VI-1967. 11 0, 22 0 (Dade Co.), Mahogany Swamp, Everglades National Park, all months except for March and May, 1964-1968. 1 0, 1 0 (Colo.), Royal Palm Hammock, Everglades National Park, 15-16-XI-1964; 28-29-IV-1965 and 4-11-1969. Georgia—2 0 (War Co.), Waycross, Laura Walker State Park, 31-VIII-1965. 1 0, 1 0 (Chatham Co.), Billy’s Island, Okefenokee Swamp, 8-XI-1965. Louisiana—1 0 (Orleans Parish), Kenner, 4-VIII-1966. 1 0 (Orleans Parish), New Orleans, 25-X-1966. Maryland—4 0, 1 0 (Anne Arundel Co.), Annapolis, 22-IX-1966. 1 0 (Wicomico Co.), Willards, 4-VIII-1968. Mississippi—2 0, 37 0 (Hancock Co.), Bay St. Louis, 26-IV, 30-VIII; 4-28 to IX-19 and 5-X-1967. North Carolina—1 0, 1 0 (Jones Co.). Trenton, 17-IX-1965. 1 0, 1 0 (Onslow Co.), Jacksonville, 9-19-IX-1965. 1 0 (Duplin Co.), Rose Hill, 4-5-VIII-1965.

DISCUSSION: Only selected references to _L. shannoni_ are given here. Forattini (1973), Martins et al. (1978) and Young (1979) provide additional references and synonyms.

There are no records of this species in Texas or in northern Mexico, north of Puebla State (Martins et al. 1978). The magnitude of this distribution may be partly due to the lack of collecting but, more likely, it reflects the absence of extensive hardwood forests, the pre-
ferred macrohabitat of this species. Future collections in east Texas may reveal its presence there. Records from Maryland and Delaware represent the known northern distribution limits of \textit{L. shannoni}.

Both sexes of \textit{L. shannoni} from the USA and the neotropics (from southern Mexico to northeastern Argentina) appear to be remarkably similar in structure. There is little or no morphological variation (Rozeboom 1944). In terms of anthropophilic behavior, however, Fairchild (1955) and others have noted that female bites humans more commonly in the USA and Mexico than elsewhere.

Rosabal and Miller (1970) collected 530 males and 233 females from tree hollows in Louisiana. No parasites were observed in 29 females dissected. Perkins (1982) dissected 414 males and 166 female \textit{L. shannoni} from 2 localities in Florida (Gulf and San Felasco Hammocks) and found acaule and garareina in 3 males (0.7%) and 15 females (9.1%). Three mites, provisionally identified as \textit{Estigmus} sp., were found attached to the abdomens of 2 female flies. The ovary of one female was parasitized by a small unidentified nematode. One female \textit{L. shannoni} collected on 17-VII-1981, Gulf Hammock, had thousands of relatively large unidentified flagellates in the midgut and foregut but not in the mouthparts. The fly was dissected 9 days after it had fed on man, but no signs of disease were subsequently observed. The midgut of a dissected male \textit{L. shannoni} from the same locality contained relatively small flagellates that were probably monoxenous.

Further studies are being made to characterize these flagellate infections.

Perkins (1982) established a laboratory colony of \textit{L. shannoni} from field-collected males in Florida (Endris et al. 1982) and provided information on its life cycle and biology.

Females normally deposit as many as 40 viable eggs without having a previous bloodmeal. Only 12 (3.4%) of 349 reared, non-blooded females failed to develop fully formed eggs by the fourth day following emergence. Autogenous females with developing eggs also took multiple bloodmeals between ovipositions, beginning 24 hours post-emergence. Laboratory hosts included man, pig, hamster, squirrel \textit{(Sciurus)}, calf, horse, opossum \textit{(Didelphis)} and dog (Endris et al. 1982). Christensen and de-Vasquez (1982), using the precipitin test for bloodmeal determination in Panama, demonstrated that \textit{L. shannoni} feeds on a variety of mammals, some birds but no reptiles. Females fed on sloths more than any other mammal species, and the authors suggested that \textit{L. shannoni} may play a role in the transmission of \textit{Leishmania braziliensis} among these reservoir hosts in Panama.

\textit{Lutzomyia shannoni} has not been found in the endemic focus of \textit{Leishmania mexicana} in Texas, but experimentally-infected \textit{L. shannoni} females (WRAIR strain 411 from Texas) transmitted the parasite to non-infected hamsters by bite (Lawyer and Young, unpublished data). Perkins (1982) reported that 26/27 lab-reared females became infected with this strain after feeding on infected hamsters. Zeledón and Alfaro (1973) found unidentified promastigotes in 4/117 (3.4%) wild-caught \textit{L. shannoni} females in Costa Rica, and later Zeledón et al. (1979) identified other flagellates recovered from this sand fly as \textit{Endotrypanum} sp. and \textit{Leishmania hertei}, both sloth parasites.

7. \textit{Lutzomyia} \textit{(Psathyromyia)} \textit{tanyopsis} Young and Perkins, n.sp., Fig. 8. \textit{Holotype} ♀ (measurements in mm). Wing length 2.28; width 0.66. Most of insect lightly pigmented, pleuron slightly paler than scutum. Head from vertex to distal end of clypeus 0.50 high; 0.38 wide. Eyes small, separated by 0.15 or by a distance = to about 0.6 facet diameters. Flagellomere I = 0.27 long, combined length of II + III = 0.53; ascoids present on all flagellomeres except last (flag. XIV), each ascoid with a long proximal spur. Labrum 0.34 long. Lengths of palpomeres: 1, 0.05; 2, 0.17; 3, 0.19; 4, 0.13; 5, 0.31; with about 12 sensilla at middle of palpomere 3. Cibarium with 4 short equidistantly-spaced horizontal teeth, middle pair inwardly directed and outer pair slanted outwardly; an irregular row of about 10 small vertical teeth present; cibarial arch complete but more diffuse in middle; pigment patch subtriangular, wider posteriorly. Pharynx 0.20 long, without spines. Pleuron with 6 upper and 4 lower episternal setae. Lengths of wing vein sections: alpha* = 0.36; beta = 0.34; delta = 0.04; gamma = 0.47. Lengths of femora, tibiae and basitarsi as follows: foreleg, 0.85, 0.83, 0.50; midleg, 0.88, 1.02, 0.54; hindleg, 0.98, 1.32, 0.63. Spermatheca spherical with basal annulations; individual ducts smooth walled, about 8X length of common duct.

**Known distribution**: USA. Arizona (Pima Co.), Fig. 20.

**Material examined**: USA. Arizona—Holotype—

\*alpha = Length of \textit{R}_2 from its junction with \textit{R}_4 to costa (see Fig. 2B).

\*beta = Length of \textit{R} from junction of \textit{R}_4 and \textit{R}_3 to junction with \textit{R}_4.

\*delta = Length of \textit{R}, extending beyond junction of \textit{R}_4 and \textit{R}_5.

\*gamma = Length of \textit{R} from origin of \textit{R} to origin of \textit{R}_{4+1} and \textit{R}_6.
type ♀ (Pima Co), Sabino Canyon, Coronado National Forest, 9-VIII-1955, light trap, G. Butler (USNM), Paratypes, 2 ♀, same data.

Discussion: We tentatively place L. tanyopsis in the subgenus *Psathyromysia* because of the long proximal spurs of the antennal ascods and the cibarial armature. Damasceno et al. (1968) differ in subgenus from those of other *Psathyromysia* females and from those of the neotropical species *L. aedilisfera* (Fairchild and Hertig), *L. drenbachi* (Causey and Damasceno) and *L. aedilisfera* (Crawford and Hertig), that also have long proximal spurs on the ascods. The undiscovered male of *L. tanyopsis* also probably will have ascodial spurs and other features that should readily associate it with the female and determine its definite placement in the genus.

The specific name, "tanyopsis," is a Greek word meaning long face or long in appearance (see Fig. 8A).

**Species Group Aragaoi Theodor**

8. *Lutromysia texana* (Dampf), Figs. 9, 16 and 17.


Psathyromys texanus: Forattini 1971:105 (listed); 1973:480 (tax.)

Known distribution: Honduras, Mexico, USA, Texas (Aransas, Bexar, Cameron, Edwards, Gillespie, Kinney, San Patricio, Uvalde and Val Verde counties). Fig. 20.


Discussion: *Lutromysia texana* is one of the largest species in North America. The wing length of females ranges from 2.55 to 2.93 mm (n = 9) and of the males from 2.30 to 2.57 mm (n = 9) (all from Texas).

The poorly preserved specimens from Honduras were tentatively identified as *L. texana*. They might actually represent *L. barretto majuscula* Young, a closely related species occurring in Colombia and Central America. It has not been established whether, or if populations of these 2 species meet in Central America. Structural differences between these taxa are slight and may reflect intraspecific variation in a single widespread species.

*Lutromysia texana* was first collected in a nest of the leaf-cutting ant, *Atta texana*, near San Antonio, Texas (Dampf 1958). Eads et al. (1965) excavated a number of *Atta* nests near Brownsville, Texas, but failed to recover larvae or resting adults of *L. texana*. Light trap catches by these authors, however, showed that adults were present throughout the year near Brownsville, but not in the Del Rio, Texas area.

Young (1972) reported that armadillo burrows serve as the usual resting sites for this species in Texas. In June 1980, one of us found 9 recently blooded females in a burrow inhabited by armadillos near Sinton, Texas, but there is no direct evidence that the flies had fed on these mammals. The flies died before their eggs were laid.

**Subgenus Helencorytonymia Barretto**

9. *Lutromysia* (*Helencorytonymia*) apache (Young and Perkins n.s., Figs. 10, 16 and 17.

Holotype ♀ (measurements in mm). Wing length 2.47; width 0.66. Head, scutum, abdominal sclerites and external genitalia inflated; rest of body pale or nearly pale. Head from vertex to tip of clypeus, 0.41 high; 0.39 wide. Eyes separated by 0.15 or by a distance = to about 8 facet diameters. Flagellomere 1. 0.27
light trap, W. Wirth. *2 allotype* (Coehsne Co.), 3.1 km SW of Portal, V-VI-1967, blacklight trap, C. Sabrosky. Other paratypes, 12 *♂, 11 ♀, same data as holotype. 2 *♀*, same data as allotype. 2 *♀* (Gila Co.), 27 km NE of Payson, Tonto Creek, 10-VIII-1978, light trap, C. Ray (UCR).

**Discussion:** Structural similarities indicate that *L. apache* is closely related to *L. saxator* and *L. oppidana*, both of which may occur with it in the USA and northern Mexico. Both sexes of *L. apache* can be separated from those of the other species by the characters given in the keys. *Lutzomyia vindicator* (Dampf), known from Mexico, differs from *L. apache* by its broader, differently shaped paramere, shorter aedeagal filaments (less than 4X the length of the ejaculatory apodeme and sperm pump), and by the wider shorter individual sperm ducts of the female. The cibarial armatures of these females are remarkably similar.

The wing length of 8 female paratypes of *L. apache* ranges from 2.42 to 2.88 mm; that of 5 male paratypes ranges from 2.37 to 2.55 mm. The specific name, "apache," refers to the tribe of American Indians living in the southwestern USA and northern Mexico.


**Known Distribution:** Mexico, USA, Colorado (Fort Collins and Larimer counties), Montana, Ravalli Co., Texas (Presidio Co.), and Washington (Adams and Whitman counties). Canada. Alberta and British Columbia, Fig. 21.

Discussion: A female of L. stearti was identified from Ukiah, Mendocino Co., California. This specimen was provisionally, but incorrectly, determined earlier as L. californica by Fairchild and Hertig (1957) and was cited as that species by Chaniotis and Anderson (1968).

Laurent (1965) collected 154 adults of L. stearti in light traps from 1 June to 10 October 1956 at Woodside, San Mateo Co., California. Chaniotis (1967) stated that in California adults of L. stearti are active for about 6 months of the year and that the fourth instar larvae diapause during the winter months.

Adults rest, and perhaps breed, in the burrows of ground squirrels, Citeulus spp., where they feed on lizards and snakes (Chaniotis and Anderson 1968).

Chaniotis (1967) reared L. stearti in the laboratory and provided data on its life cycle, feeding and mating behavior, and other habits. Ayala and Lee (1970) observed sporozoites of Plasmodium mexicanum, a saurian malaria, in wild-caught L. stearti after they were experimentally infected in the laboratory.

12. Lutzomyia (Helocerytomyia) vexator (Coquillet), Figs. 8 and 13.


Phlebotomus vexator occidentalis Fairchild and Hertig 1957:334 (♂, ♀, Alturas trap station, Modoc Co., California and Topaz Lake, Mono Co., California). Fairchild and Harwood...


Discussion: Fairchild and Herrig (1957) studied males and females of L. vexator from California and concluded that they differed
subspecificaly from specimens collected on Plummer’s Island, Maryland, the type-locality. They stated that the California specimens, which they named L. vexator occidentis, had the following distinguishing characteristics: “Male with gonocoxal filaments shorter, less than 4 times length of uropod; style shorter and stouter, the two subapical spines farther apart and the most basal one of this pair closer to the basal spine than in vexator. Basal tuft of subulate of 3 setae. Female with spermatovisceral shorter, more slender, less than twice as long as stem of genital fork. Cibarium narrower, with but 4 horizontal teeth, lacking the sublateral shorter teeth found in vexator.”

Chaniotis and Anderson (1964, 1967) redescribed L. v. occidentis from California specimens, noting that the aedeagal filaments of 10 males were 4 times the length of the ejaculatory apodeme and spermatophore, and that the basal gonocoxal tuft consisted of 4 or 5 long hair-like setae. The length of the aedeagal filaments of Florida males that were examined in this study varied from 3.5 times to 4.6 times the length of the ejaculatory apodeme and spermatophore, and the gonocoxal tuft consisted of 4 or 5 setae. This range of measurements encompasses that observed for all other males examined (including California and Washington State specimens) except that one male from Plummer’s Island had aedeagal filaments 4.9 times the length of the ejaculatory apodeme and spermatophore. The length of the gonostylus and presence of a subulate of teeth in the female cibarium are not diagnostic. Of 7 females examined from the same population in Texas, 2 had sublateral teeth while the other 5 lacked them. The length of the gonostylus of 16 males from Washington State ranged from 0.21 to 0.24 mm and from 0.24 to 0.26 for 3 males from Maryland. In view of this variation and of our inability to detect clear-cut, consistent differences among specimens from various localities, L. vexator is regarded as a morphologic species that has a widespread geographic distribution in North America.

*Lutzomyia vexator* is easily reared in the laboratory. Marshall Hertig maintained a closed colony from 1932 to 1936 at Harvard University from specimens collected at Plummer’s Island, Maryland. Chaniotis and Anderson (1964) and Chaniotis (1968) successfully reared *L. vexator* and provided information on its biology under laboratory conditions and in the field (Chaniotis and Anderson 1968). A laboratory colony is presently being maintained at the University of Colorado (Endris et al. 1982). Many and colleagues (1968-75) published several articles on sand flies (mostly *L. vexator*) and associated parasites in California. There, the sand flies rest in ground squirrel burrows that are also occupied by lizards, snakes and other cold-blooded vertebrates which serve as hosts for these insects.

Sporozoites of *Plasmodium mexicanum* a sau- rian malaria, were observed in experimentally-infected sand flies (mostly *L. vexator*) from California. These sporozoites were infective to *Sceloporus* lizards when injected intraperitoneally. Transmission was not demonstrated by sand fly bite and no natural infections were found in the presumed vector.

During the past 5 years in Florida, over 600 laboratory-reared females of *L. vexator* that had previously fed on *Plasmodium floridense-infected* anoles (*Anolis carolinensis*) were dissected. Oocysts were seen in over 50% of the flies that were dissected 4-20 days after the infecting bloodmeal, but no sporozoites were detected.

In addition to these preliminary observations, it is noted that naturally infected *Anolis* have been found in several Florida localities where phlebotomine sand flies do not occur. Although hardly conclusive, these observations suggest that *P. floridense* is not transmitted to *Anolis* and fence lizards (Sceloporus) in Florida by phlebotomines.

**SUBGENUS MICROPYGOMYIA** Barretto


**KNOWN DISTRIBUTION:** USA. Arizona—(Pinal Co.). California (Imperial, Inyo, Kern, Lassen, Monterey, Riverside, San Diego and Yolo counties). Texas (Presidio and Val Verde counties). Washington (Adams Co.), Fig. 23.

Discussion: Fairchild and Hertig (1957) pointed out that L. chiapensis (Dampf) are very similar in structure, differing only in the relative lengths of the female sperm ducts and in details of the male terminalia. Now that additional material is available, there is a strong possibility that these differences reflect geographic variation of a single widespread species that occurs from Panama to northwest USA. We have not examined specimens from Mexico, however, and will therefore continue to regard these species as distinct.

Apart from the longer common sperm duct of the L. chiapensis female there are no other structural differences between it and L. californica. There is considerable variation in the number of horizontal teeth in the female cibarium. Chaniotis and Anderson (1968) counted 16–34 such teeth in 10 or more females from California. The Texas female, examined by us, has 20 teeth; 13 females from Othello, Washington have 16–25 teeth. Females referable to L. chiapensis show similar variation. The female holotype of L. chiapensis from Chiapas State, Mexico, has 25 horizontal teeth (Dampf 1947). Specimens from Panama (n = 5), Costa Rica (n = 1) and El Salvador (n = 1) have 20–24, 20 and 28 teeth respectively.

The males of L. californica and L. chiapensis are distinguished by the shape and setation of the gonocoxites. Unlike L. californica, Lutyzmyia chiapensis has a gonocoxite that is noticeably widened basally and has numerous setae distal to the basal tuft.

In California, females of L. californica feed on lizards and snakes, probably within mammal burrows which also serve as resting sites for the flies (Chaniotis and Anderson 1968). These authors collected L. californica in Yolo Co., California, in low numbers from June 24 to October 12, 1965, and suggested that there are 3 generations per year in that area.

14. Lutyzmyia (Microptepymia) cubensis (Fairchild and Trapido), Fig. 15.


Known distribution: Cuba. USA. Florida (Monroe Co.), Fig. 23.


Discussion: Lutyzmyia cubensis is a small sand fly that successfully colonized some of the Florida Keys presumably from West Indian stock. Its close relatives, L. coyennensis (Flock and Abonnenc) and allies, feed mostly on lizards.

CONCLUDING REMARKS

Much more information on sand flies in North America is needed to understand the extent of their geographic ranges, biometrics and disease relationships. With few exceptions, most observations and collections of these flies were made incidental to other insect studies. The phlebotomine fauna of the western USA is richer than that of the eastern states and it is there that additional species are likely to be discovered and where studies on the known species are especially needed.

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References Cited


Fig. 2. *Luizomyia cruciata* (Coquillett). A. Male terminalia, lateral view; B. Female wing; C. Male wing; D. End of female abdomen showing papillate lobe of tergite 8; scale in mm; E. Spermathecae and genital fork; F. Male head; G. Female cibarial; H. Male flagellomere II; I. Female flagellomere II; J. Female head. Drawn from Florida specimens at same scale as comparable structures in Fig. 4.
Fig. 3. *Lutzomyia diabolica* (Hall). A. Male head; B. Male terminalia; C. Female wing; D. Male wing; E. Spermathecae and genital fork; F. Female charum; G. Female head; H. Male flagellomere II; I. Female flagellomere II. Drawn from Texas specimens at same scale as comparable structures in Fig. 4.
Fig. 4. Lutzomyia zerephala Young, Brenner and Wargo. A. Female flagellomere II; B. Female head; C. Male flagellomere II; D. Male head; E. and F. Different views of male parameres; G. Male aedeagal filaments, ejaculatory apodeme and sperm pump drawn at same scale as Fig. 4I; H. Spermatheca drawn in Canada balsam; I. Spermathecae and genital fork drawn in phenol; J. Female wing; K. Male wing; L. Male terminalia; M. Female cibarium; same scale as Fig. 4N; N. Female cibarium. All figures after Young et al. (1985), except Fig. 4M, from California specimens. Scale in mm.
Fig. 5. *Lutzomyia aquilonia* (Fairchild and Harwood). A. Male terminalia; B. Female wing; C. Male wing; D. Spermathecae and genital fork; E. Spermatheca of allotype; F. Male aedeagal filaments, ejaculatory apodeme and sperm pump; G. Female head; H. Male head; I. Female flagellomere II; J. Male flagellomere II; K. Female cibarium. Drawn from Washington State specimens at same scale as comparable structures in Fig. 4.
Fig. 6. *Luizomyia androphora* (Addis). A. Male terminalia; B. Male aedeagal filaments, ejaculatory apodeme and sperm pump; C. Female wing; D. Male wing; E. Male flagellomere II; F. Male head; G. Female flagellomere II; H. Female head; I. Spermathecae showing bubble-like evaginations and genital fork; J. Female clavarium. Drawn from Texas specimens at same scale as comparable structures in Fig. 4.
Fig. 7. *Luettia myia shannoni* (Dyar). A. Male terminalia; B. Male aedeagal filaments, ejaculatory apodeme and sperm pump; C. Female flagellomere II; D. Female wing; E. Male wing; F. Female head; G. Spermathecae (thin outer envelope surrounding each spermatheca not shown) and genital fork; H. Male flagellomere II; I. Male head; J. Female cirri. Drawn from Florida specimens at same scale as comparable structures in Fig. 4.
Fig. 8. *Lutzomyia tanygnia* Young and Perkins n.sp. (SA-E holotype) and *Lutzomyia vexator* (Coquillett) from Pt. Ord, California (8F). A. Female head; B. Female cibarium; C. Female flagellomere II; D. Spermathecae and genital fork; E. Female wing; F. Spermathecae and genital fork. All structures drawn at same scale as those in Fig. 4.
Fig. 9. *Lutzomyia tznana* (Dampf). A. Male terminalia; B. Male aedesal filaments, ejaculatory apodeme and sperm pump; C. Spermathecae and genital fork; D. Male flagellomere II; E. Female flagellomere II; F. Female cibarium. Drawn from Texas specimens at same scale as comparable structures in Fig. 4. Also see Figs. 16G, H, and 17G, H.
Fig. 10. *Lutzomyia apache* Young and Perkins n.sp. A. Male terminalia; B. Male aedeagal filaments, ejaculatory apodeme and sperm pump; C. Male flagellomere II; D. Female flagellomere II; E. Female cibarium; F. Spermatheca (only one is shown) and genital fork. Drawn from Springerville, Arizona, specimens at same scale as comparable structures in Fig. 4. Also see Figs. 16E, F and 17E, F.
Fig. 11. *Lutzomyia affinis* (Dampf). A. Male terminalia; B. Male flagellomere II; C. Female flagellomere II; D. Female cibarium; E. Spermathecae and genital fork; F. Tip of aedeagal filament; G. Aedeagal filaments, ejaculatory apodeme and sperm pump. Drawn from Mexico specimens at same scale as comparable structures in Fig. 4. Also see Figs. 16A, B and 17A, B.
Fig. 12. *Lutzomyia stewarti* (Mangabeira and Galindo). A. Male aedeagal filaments, ejaculatory apodeme and sperm pump; B. Male terminalia; C. Spermathecae and genital fork; D. Male flagellomere II; E. Female flagellomere II; F. Female cibarium. Drawn from California specimens at same scale as comparable structures in Fig. 4. Also see Figs. 16C, D and 17C, D.
Fig. 13. *Lutzomyia vesator* (Coquillett). A. Male terminalia; B. Spermathecae and genital fork; C. Female cibarium; D. Male aedeagal filaments, ejaculatory apodeme and sperm pump with one filament tip enlarged; E. Female wing; F. Male wing; G. Male flagellomere II; H. Male head; I. Male flagellomere II; J. Female flagellomere II. Drawn from Florida specimens at same scale as comparable structures in Fig. 4. Also see Fig. 8F.
Fig. 14. *Lutzomyia californica* (Fairchild and Hertig). A. Male terminalia; B. Spermathecae and genital fork; C. Male aedeagal filaments, ejaculatory apodeme and sperm pump; D. Female wing; E. Male wing; F. Male cibarium; G. Female head; H. Male head; I. Female cibarium; J. Female flagellomere II; K. Male flagellomere II. Drawn from California specimens at same scale as comparable structures in Fig. 4.
Fig. 15. *Lutzomyia cubensis* (Fairchild and Trapido). A. Male terminalia; B. Female wing; C. Male wing; D. Male flagellomere II; E. Female flagellomere II; F. Female cibarium and pharynx; G. Female head; H. Male head; I. Spermathecae and genital fork. Drawn from Florida specimens at same scale as comparable structures in Fig. 4.
Fig. 16. Male and female heads of *Lutzomyia oppidana* (A and B); *L. stewarti* (C and D); *L. apache* (E and F), and *L. texana* (G and H). All drawn at same scale as heads in Fig. 4.
Fig. 17. Male and female wings of *Lutzomyia oppidana* (A and B); *L. stewarti* (C and D); *L. apache* (E and F); *L. texana* (G and H). All drawn at same scale as wings in Fig. 4.
Fig. 18. Distribution of *Lutzomyia cruciata* and *L. diabolica* in North America.

Fig. 19. Distribution of *Lutzomyia xerophila*, *L. aquilonia*, *L. anthophora* and *L. shannoni* in North America.
Fig. 20. Distribution of *Lutzomyia tanyopsis*, *L. texana* and *L. apache* in North America.

Fig. 21. Distribution of *Lutzomyia oppidaana* and *L. stewarti* in North America.
Fig. 22. Distribution of Lutomyia vexator in North America.

Fig. 23. Distribution of Lutomyia californica and L. cubensis in North America.