THE UNIVERSITY OF MANITOBA

THE BIOLOGY AND SYSTEMATICS OF *AEDES CAMPESTRIS* DYAR AND KNAB (DIPTERA:CULICIDAE) AND RELATED SPECIES IN MANITOBA AND SASKATCHEWAN

by

PENSOOK TAUTHONG

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PENSOOK TAUTHONG

A dissertation submitted to the Faculty of Graduate Studies of the University of Manitoba in partial fulfillment of the requirements of the degree of

DOCTOR OF PHILOSOPHY

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ABSTRACT

Voltinism was determined in various populations of *Aedes campestris* Dyar and Knab from different locations. The Winnipeg population was 100% multivoltine, whereas the Churchill population was 64% multivoltine, 36% univoltine. Saskatoon and Estevan populations were 99% multivoltine, 1% univoltine. A laboratory colony of *A. campestris* was successfully maintained in a 120-x 120-x 210-cm cage for 3 generations. The optimum temperature for development and survival of *A. campestris* in the laboratory was 23°C. The time required for larval and pupal development was 9 and 3 days respectively at 23°C. Ovarian development of blood-fed females was also studied at 23°C, and females were able to complete follicular development within 3-4 days. Variation in autogeny was observed in populations from different geographic regions. In the Churchill population, 45% of the females were able to develop some autogenous eggs 15-20 days after emergence. In *A. campestris* from Saskatoon, 13.9% of the females developed a few autogenous eggs. There was no significant difference between the number of eggs laid by females fed 1, 2, or 3 blood meals in the Saskatoon or Estevan populations. The Churchill females fed 3 blood meals produced more eggs than those fed 1 blood meal.

At 23°C the critical period for induction of egg diapause in the Saskatoon population occurred between 13L:11D and 14L:10D. Egg diapause in the Saskatoon population was obtained after 10-14 days of exposure to a short photoperiod (<14L:10D). The greatest percentage of diapause occurred after 30 days. Long photoperiod (16L:8D) terminated egg diapause at 23°C in the Churchill population and the percentage of

termination increased significantly at 30°C.

The larval and adult stages of *A. campestris* and the related species, *A. dorsalis* (Meigen) and *A. mediolineata* Ludlow, are described. This study confirmed the separate species status of *A. melanimon* from *A. dorsalis* and reveals an older name than *A. melanimon*, namely *mediolineata* Ludlow 1907 which was previously a synonym of *A. dorsalis*. The key for species and the comparisons of various characters of first and fourth instar larvae, and male and female adults are listed to point out the reliable characters for separating each species. The distribution of the three species in North America is illustrated in a map.

TABLE OF CONTENTS

	Page
ABSTRACT	ii
TABLE OF CONTENTS	iv
LIST OF TABLES	vi
LIST OF FIGURES	vii
ACKNOWLEDGEMENTS	ix
INTRODUCTION	1
PERTINENT LITERATURE	4
METHODS AND MATERIALS	15
GENERAL METHODS	15
COLONIZATION TECHNIQUES	18
INDUCTION, MAINTENANCE, AND TERMINATION OF EGG DIAPAUSE	20
RESULTS	23
DETERMINATION OF VOLTINISM IN DIFFERENT POPULATIONS OF *A. CAMPESTRIS*	23
EFFECT OF TEMPERATURE ON LARVAL, PUPAL, AND ADULT DEVELOPMENT	23
MATURATION OF OVARIAN FOLLICLES	27
AUTOGENY IN *A. CAMPESTRIS*	31
MATING SUCCESS IN *A. CAMPESTRIS*	36
EFFECT OF THE NUMBER OF BLOOD MEALS ON FEMALE FECUNDITY	36
EFFECT OF PHOTOPERIOD ON THE INDUCTION OF EGG DIAPAUSE	39
EFFECT OF PHOTOPERIOD AND TEMPERATURE ON THE TERMINATION OF EGG DIAPAUSE	39

Page

SPECIES DESCRIPTIONS 44

DESCRIPTION OF *A. CAMPESTRIS*. 44

DESCRIPTION OF *A. DORSALIS*. 66

DESCRIPTION OF *A. MEDIOLINEATA*. 74

COMPARISON OF LARVAL AND ADULT CHARACTERS OF *A. CAMPESTRIS*, *A. DORSALIS* AND *A. MEDIOLINEATA*. 80

KEY TO SPECIES 87

DISCUSSION 90

SUMMARY. 96

REFERENCES CITED 98

LIST OF TABLES

Page

Table

1. Determination of voltinism in different populations of *A. campestris* 24
2. Effect of temperature on development and survival of *A. campestris* 25
3. Rate of follicular development in blood-fed females of *A. campestris* at 23°C 32
4. Autogenous egg development in *A. campestris*, from Churchill and Saskatoon reared at 23°C and photoperiod 16L:8D. 34
5. Mating success of *A. campestris* in 120-x 120-x 210-cm cage. 37
6. Effect of the number of blood meals on the fecundity of *A. campestris* at 23°C 38
7. Effect of photoperiod on the induction of diapause in multivoltine eggs of *A. campestris* from Saskatoon, at 23°C 40
8. Effect of long photoperiod and temperatures on termination of diapause in eggs of *A. campestris* from Churchill 43
9. Comparison of larval and adult characters of *A. campestris*, *A. dorsalis* and *A. mediolineata*. 81

LIST OF FIGURES

FIGURE Page

1. Rate of growth of *A. campestris* larvae at 5 temperatures 27
2. Stages of follicular maturation in female imagos of *A. campestris* 30
3. Stages of follicular maturation at 1, 2, and 3 days after a blood meal in female imagos of *A. campestris* reared at 23°C 33
4. A group of follicles of a facultatively autogenous female of *A. campestris* from Saskatoon, Saskatchewan. . 35
5. Effect of different photoperiods on the induction of egg diapause in *A. campestris* (Saskatoon population) at 23° C 42
6. Parts of the thorax and pleural chaetotaxy of an adult *Aedes* mosquito 46
7. Dorsal view of the thorax of the adult females of *A. mediolineata*, *A. campestris*, and *A. dorsalis* 47
8. Dorsal view of the abdomen of the adult females of *A. mediolineata*, *A. campestris*, and *A. dorsalis*. . . . 49
9. Lateral view of the claw of an adult female (foreleg). 50
10. Claws of adult females of *A. mediolineata*, *A. campestris* and *A. dorsalis* 51
11. Pattern of the wing scales in adult females of *A. mediolineata*, *A. campestris*, and *A. dorsalis* 53

FIGURE Page

12. Anterior and posterior claws of adult males of *A. mediolineata, A. campestris,* and *A. dorsalis* . . . 54

13. Diagram of male terminalia of an *Aedes* mosquito. 55

14. Male terminalia of *A. mediolineata, A. campestris,* and *A. dorsalis* 56

15. Anatomical features of the 4th instar larva of an *Aedes* mosquito. 58

16. Dorsal view of the head and thorax of the 4th instar larvae of *A. mediolineata, A. campestris,* and *A. dorsalis* 59

17. Lateral aspect of abdominal segments VIII-X of the 4th instar larvae of *A. mediolineata, A. campestris,* and *A. dorsalis* 61

18. First instar larvae of *A. campestris* and *A. dorsalis*. . 62

19. Geographical distribution of *A. campestris, A. dorsalis,* and *A. mediolineata* in North America (North of Mexico) 64

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INTRODUCTION

Aedes campestris Dyar and Knab, and the closely related mosquitoes viz. *Aedes dorsalis* (Meigen) and *Aedes mediolineata* Ludlow (= *A. melanimon* Dyar.), are considered pests of man and animals throughout the Canadian West and the central and western United States. In both Canada and the United States, *A. campestris* and *A. dorsalis* are known as spring species; the females are pests in late spring and early summer. *A. dorsalis* and *A. campestris* have been found carrying the virus of western equine encephalitis (WEE) in Canada (McLintock *et al.*, 1970) and the United States (Ferguson, 1954). In the United States, *A. mediolineata* has been found carrying the WEE (Ferguson, 1954), St. Louis Encephalitis Virus (Reeves *et al.*, 1962), and probably California Encephalitis Virus (Richards, 1956). The time of peak abundance of *A. mediolineata* in Canada is not known, but in the United States it is known to breed in inland irrigated pastures and other irrigated crops throughout the summer (Telford, 1958; Chapman, 1960).

The geographical distribution in North America of these three species is probably fairly similar. Of the three, the most widely recognized species is *A. dorsalis*. It is recorded from 6 provinces in Canada and from 32 states in the United States (Carpenter and LaCasse, 1955; Richards *et al.*, 1956; Silverly, 1972). *A. campestris* is recorded from 7 provinces and 17 states (Carpenter and LaCasse, 1955; Chapman, 1966). *A. mediolineata* is recorded under the name of *A. melanimon* from 2 provinces, Alberta and Saskatchewan, and 12 states (Richards, 1956; Richards *et al.*, 1956; Burgess, 1957; Holmberg and Trofimenkoff, 1968; Gjullin and Eddy, 1972). From the present study, we know *A. mediolineata*

also occurs in British Columbia.

A. dorsalis (Meigen) 1830 was the first of these three species to be described. *A. campestris* Dyar and Knab 1907 was described later, and is recognized by culicidologists to be different from *dorsalis* in several important ways. *A. mediolineata* Ludlow 1907 is also different from *dorsalis*, and some of these differences were listed by Dyar (1924). The characters used in keys for the separation of these three species are inadequate for clear separation of larvae or adults. A reexamination of these characters has been long overdue, and an attempt is made here to emphasize the reliable characters and to delineate each species as completely as possible. Collections of larvae and adults of *A. campestris* and *A. dorsalis* were made in various locations in Manitoba and Saskatchewan. Specimens of all three species were obtained from the Canadian National Collection in Ottawa, from the United States National Museum, and from the personal collection of several researchers in both countries.

Although *A. campestris* is a common species in North America, especially in Saskatchewan, details of its biology and behaviour are not well understood. The colonization and maintenance of a laboratory colony has not previously been reported. This dissertation describes a technique developed for the successful colonization of *A. campestris*. The anatomical studies of larvae and adults of *A. campestris*, in comparison to larvae and adults of the related species *A. dorsalis* and *A. mediolineata*, are also presented. The characteristics of both instar I larvae and instar IV larvae are illustrated. Various aspects of the biology of *A. campestris* were studied. Laboratory experiments were conducted on the rate of larval development at different temperatures, adult female fecundity, autogeny in different populations, egg-follicle development,

and factors that induce and terminate egg diapause.

PERTINENT LITERAUTRE

Mosquitoes of Western Canada

The study of the mosquitoes of Western Canada was begun as early as 1866 by Lord (see Curtis, 1967). Later in 1922 Dyar published his paper on the mosquitoes of Canada after his visit to British Columbia (Dyar, 1922). The work of Hearle (1926) on the mosquitoes of the lower Fraser Valley and Curtis (1967) on the mosquitoes of British Columbia served as the basis for ecological studies and mosquito control in British Columbia.

The study of the mosquitoes of Saskatchewan began in 1907 when Knab (1908) published his observations on seven species of mosquitoes from Oxbow, Saskatchewan. His report included an extensive account of the bionomics of *Aedes* species, especially *A. spencerii* (Theobald). Another brief report on Saskatchewan mosquitoes was provided by Cameron (1918). Hearle (1929) published on the biology of mosquitoes of the Canadian prairies in which the life history of *Aedes flavescens* Müller was described.

An urgent need for further study of the prairie mosquitoes occurred following the 1941 outbreak of equine encephalitis in Saskatchewan. Rempel prepared a guide to the mosquito larvae of western Canada (1950), and subsequently a guide to the adults of Saskatchewan (1953), in which he illustrated the important taxonomic characters, and included the species descriptions and a brief note on their biology and distribution.

Mosquitoes of Manitoba were first studied by Knab (1908), who reported 8 species from Winnipeg. McLintock (1944) reported 22 species from Winnipeg, and published on their seasonal distribution, and relative abundance. Studies on biting flies at Churchill, Manitoba were carried

out by Twinn *et al.* (1948), and Hocking *et al.* (1950).

Systematics of *A. campestris* and related species

The adult male and female of *A. campestris* were first described by Dyar and Knab (1907). Rempel (1950, 1953) and Carpenter and LaCasse (1955) also described the adults and the fourth-instar larva of this species. The first-instar larva of *A. melanimon* has been described by Dodge (1966). Bohart (1954), Price (1960), and Dodge (1966) prepared keys for the identification of the first-instar larvae of *A. dorsalis* and other common mosquitoes of North America, but *A. campestris* was not included.

Prior to 1955, *A. melanimon* had been considered by some authors to be a "race" of *A. dorsalis* (Freeborn, 1926) and by others a synonym of *A. dorsalis* (Matheson, 1929; Freeborn and Bohart, 1951). More recently *A. melanimon* has been treated as a separate species (Barr, 1955; Carpenter and LaCasse, 1955; Bohart, 1956; and Richards, 1956). The present study confirms the separate species status of *A. melanimon*, and reveals an older name than *melanimon* for this species, namely *mediolineata* Ludlow 1907, previously a synonym of *A. dorsalis*.

Characters that separate *A. campestris*, *A. dorsalis* and *A. mediolineata* have been noted by several workers. Generally, *A. campestris* is reported to be larger and darker than *A. dorsalis*. Although quite similar, the male terminalia of *A. campestris* can be separated from *A. dorsalis*. The tarsal claws are reported to be the most reliable character for separating the females (Rempel, 1953; Vockeroth, 1954; Richards, 1956; Barr, 1958; Harmston and Lawson, 1967). Vockeroth (1954) stated that the claw of the metathoracic leg of the male adult could be used for identification. Although the wing scale

pattern on vein R_{4+5} has been one of the characters used for identification by several authors (Matheson, 1929; McLintock, 1944; Rempel, 1953; Chapman, 1966; Carpenter and LaCasse, 1955; Harmston and Lawson, 1967), Barr (1958) and Ross (1947) do not consider this to be a reliable character.

The identification of *Aedes* eggs based on the chorionic pattern, and the shape and size of eggs has been reported by many authors (Horsfall and Craig, 1956; Craig and Horsfall, 1958, 1960; Ross and Horsfall, 1965; Myers, 1967; Kalpage and Brust, 1968; Horsfall *et al.*, 1970; Horsfall and Voorhees, 1972 etc.). Myers (1967) described the egg size, shape, and chorionic pattern of *A. campestris*, *A. dorsalis*, and *A. melanimon* as well as 20 other species from California and Nevada. The chorionic detail of the egg of *A. campestris* is very close to that of *A. dorsalis* and *A. melanimon*. The size of an *A. melanimon* egg is relatively small (498-597 μ in length) compared to *A. dorsalis* (660 - 759 μ) and *A. campestris* (597-763 μ) (Myers, 1967). The size of eggs varies considerably within a species.

According to Kalpage and Brust (1968) there are 2 types of *A. dorsalis* eggs in populations in Manitoba; the length of eggs was 553-643 μ for one race and 655-732 μ for the other, and the length of *A. campestris* eggs was 681-822 μ. They suggested that there are two forms of adults, or two races in *A. dorsalis*. It is possible that one of their races of *A. dorsalis* could be *A. mediolineata*, however, this has not been confirmed.

Biology of *A. campestris*

The biology of mosquitoes common to Western Canada can be found in

Hearle (1929), Rempel (1950, 1953), Carpenter and LaCasse (1955), Horsfall (1955), Barr (1958), Clements (1963), and Happold (1965).

Prior to the present study, the following was known about *Aedes campestris*. Larvae may be found around Winnipeg from May 10 - June 15 in temporary and semipermanent pools formed by melting snow or heavy rains (McLintock, 1944). The pools occur in open areas, often near refuse dumps, and the water temperature during larval development may vary from 12-30°C. At Churchill, Manitoba, Twinn *et al.* (1948) found early instar larvae of *A. campestris* in shallow pools among marsh grasses and dwarf willow and birch on June 18. Larvae also occurred in a burnt-over forest region which was largely open. The bottom of the pool consisted of soft black muck, with a yellow surface. The salinity of the pool was 368 ppm and the pH was 8.3. Hocking *et al.* (1950) reported the pupal period in *A. campestris* at Churchill to be 2-7 days, and a mean of 4.8. Adults were abundant from June 30-July 23. Swarming of adults was observed about 8.30 p.m., over a forest clearing at the height of 6 - 12 ft (Hocking *et al.*, 1950). In Utah, adults of *A. campestris* are known to fly as far as 10 miles (Rees, 1943). Mail (1934) reported egg laying 4 days after a blood meal, whereas Hocking *et al.* (1950) observed that females did not oviposit until 15-16 days after feeding. Fully embryonated eggs have been stored for periods up to 20 months at 0-10°C, with more than 25% survival of the embryos (Mail, 1934). In nature, eggs of *A. campestris* probably hatch before those of *A. dorsalis*, since larvae and adults of *A. campestris* usually appear first (Barr, 1958). Only one generation per year was observed by McLintock (1944), Rempel (1950), Barr (1958), and Chapman (1966).

Laboratory Colonization of *Aedes dorsalis*

Blakeslee *et al.* provided a preliminary report on the colonization of *A. dorsalis* in a 180-x 120-x 210-cm cage. However, due to the limitation of large space requirements, an attempt was made to establish a strain requiring less space. The subcolony was established after 9 generations in a 120-x 60-x 60-cm cage and through 6 generations in a 60-x 60-x 60-cm cage. Egg-hatch rates of the larger-cage subcolony showed a steady increase with each generation whereas the smaller-cage subcolony showed an initially high rate, then dropped somewhat, and finally began to increase (Grimstad *et al.*, 1970). Taylor and Brust (1974) also reported the successful colonization of *A. dorsalis* and *A. vexans* (Meigen) in 120-x 120-x 210-cm cage. *A. vexans* mated more readily in the presence of *A. dorsalis*.

The Effect of Temperature on Larval Development

The effects of temperature on pre- and post- adult development of various mosquito species have been studied by Bar-Zeev (1958) in *Aedes aegypti* (Linnaeus); Brust (1967) in *A. vexans*, *A. nigromaculis* (Ludlow), and *Culiseta inornata* (Williston); Hanec and Brust (1967) in *Culiseta inornata;* Trpis and Horsfall (1969) in *A. sticticus* (Meigen); Trpis and Shemanchuk (1969, 1970) in *A. flavescens* and *A. vexans;* and recent work by Smelton (1973) on 8 mosquito species. None of these studies involved *A. campestris*.

Follicular Development

The stage of development of the ovaries has been classified into 5 different categories by Christophers (1911) and modified by Clements

(1963). The rate of development of the oocytes from the resting stage to maturity in a blood-fed female is controlled by several factors. One of these is temperature which acts indirectly by affecting the rate of digestion of blood, and directly on the growth of the oocyte (Clements, 1963). Another factor is genetics, which may be manifested by inter- or intra-species differences. *Anopheles gambiae* Giles oviposited 2 days after blood feeding at temperatures above 23.3° C while *Anopheles funestus* Giles required 3 days at 24.7° C (Gillies, 1953). *Anopheles vagus* Dönitz females from Assam were able to lay eggs 24 hours after taking a blood meal (Muirhead-Thomson, 1951). Females of *Anopheles maculipennis* Meigen from Stalingrad completed 12 gonotrophic cycles in 42 days in Southern Russia whereas in Moscow it took about 79 days to complete the same number of cycles (Detinova, 1962). Spielman (1957) found that ovarian development in autogenous females of *Culex pipiens* Linnaeus was suspended for 10 days or more at temperature of 4.4° C but development was resumed at normal temperature. According to Clements (1963) the gonotrophic cycles in mosquitoes varied greatly under fluctuating temperature, especially in temperate regions, but in tropical regions they were fairly constant, being only 2 days in most species.

Autogeny

The term autogeny was used by Roubaud (1929) to indicate the ability of unfed females to develop eggs. Spielman (1957) described autogeny as the absence of a developmental diapause condition of the ovary. Chapman (1962) characterized autogenous females as those that were capable of developing fully formed eggs without a blood meal or

external source of protein.

The first observation on autogeny was made on *Culex pipiens* by Theobald (1901). Roubaud (1933) found that both autogenous and anautogenous populations occurred within the same species, *Culex pipiens*. Spielman (1971) evaluated the environmental factors that influenced the relative abundance of these 2 forms in nature. He found that autogenous mosquitoes were most numerous in larval developmental sites that were enclosed, whereas anautogenous mosquitoes were most numerous from more open bodies of water. DeBoissezon (1933) suggested that autogeny in *Culex pipiens* depended on the larval diet, not on racial or genetic characteristics. The difference between these 2 populations was shown to be due to differences in the hormonal control of ovary growth (Clements, 1956; Larsen, 1958; Larsen and Bodenstein, 1959). However, in *Aedes aegypti* and *Anopheles maculipennis*, the larval nutrition is in some way connected with the appearance of autogeny in a population (Clements, 1963).

Geographical variation also influences the occurrence of autogeny in field populations. O'Meara and Evans (1973) have shown that the capacity of females to produce autogenous eggs increased from northern to southern populations of *Aedes taeniorhynchus* (Wiedemann) in Florida.

Other factors which influence autogeny in *Aedes communis* (DeGeer) have been studied by various workers. According to Beckel (1954) proteins required for autogenous ovarian development are obtained from the fat body and larval abdominal muscles of *A. communis*, whereas in the same species some populations are reported to histolyse their indirect flight muscles (Hocking, 1954). Recent work by Ellis and Brust (1973) showed that *A. communis* from Canada and the United States consisted of

3 sibling species, *A. communis*, *A. nevadensis* (Chapman and Barr), and *A churchillensis* sp. n. An autogeny survey revealed that the female adults of *A. communis* and *A. nevadensis* are normally obligatorily anautogenous whereas *A. churchillensis* are normally obligatorily autogenous.

Autogeny was also reported in *A. impiger* (Walker) and *A. nigripes* Zetterstedt (Corbet, 1964; 1967); in *A. rempeli* Vockeroth (Smith and Brust, 1970); in *A. caspius* Pallas, *A. detritus* (Haliday), *Culex modestus* Ficalbi and *C. pusillus* Macquart (Chinaev, 1964); in *A. concolor* Taylor, and *Tripteroides tasmaniensis* (Strickland)(Dobrotworsky, 1954); in *A. togoi* (Theobald) (Lien, 1960; Laurence, 1964); in *Culex tarsalis* Coquillett (Bellamy and Kardos, 1958; Moore, 1963); in *Culex peus* Speiser and *Culiseta inornata* (Williston) (Washino and Shad-del, 1969); in *Toxorhynchites* Theob. (Chapman, 1962); in *Wyeomyia smithii* (Coquillett) (Price, 1958); and also in the subfamily Anophelinae (Detinova, 1962).

Autogeny appeared to be common in *A. campestris*, but less common in *A. dorsalis* and *A. melanimon* (Chapman, 1962). The autogeny investigations were based on numbers of eggs laid by females. It took 10 days for autogenous *A. campestris* to deposit the first egg, 11 days for *A. melanimon*, and 14 to 15 days for *A. dorsalis*.

Fecundity

The number of eggs laid by a single female during one gonotrophic cycle varies greatly between species. *Aedes* species usually lay 100-150 eggs, as in *Aedes aegypti* and *A. polynesiensis* Marks (Woke *et al.*, 1956; Ingram, 1954) but *A. detritus* (Haliday) lays up to 260 (Marshall, 1938).

There are several factors that affect the number of eggs laid by mosquitoes. Jalil (1974) indicated that the factors that affect the number of eggs laid by *A. triseriatus* (Say) are blood source, blood amount, body size, body weight, mating, and age of the mosquito. More eggs were laid by females when fed on warm-blooded than on cold-blooded animals. The number of eggs laid in a batch by *A. aegypti* and *A. triseriatus* shows a positive correlation with the amount of blood ingested (Roy, 1936; Woke *et al.*, 1956; Colless and Chellapah, 1960; Jalil, 1974). However, this correlation only occurs with medium-sized and small blood meals and there is no increase in egg production after 3 mg of blood have been ingested (Clements, 1963). Meola and Lea (1972) demonstrated that when ovaries of *Aedes* spp. had retained sufficient eggs, another blood meal failed to initiate the development of more eggs.

In *A. triseriatus*, the number of eggs laid showed a positive correlation with the amount of blood ingested from 0.6 mg to 3.0 mg (Jalil, 1974) and 1 to 5 mg were required in *A. hexodontus* Dyar (Barlow, 1955). It was found that larger females of *A. hexodontus* ingested more blood than smaller females (Barlow, 1955). Females of *A. aegypti* derived from well-nourished larvae ingested more than twice as much blood as females reared under starvation conditions (Bar-Zeev, 1957). It has also been shown that the size of the female has a positive correlation with number of ovarioles in the ovaries. Colless and Chellapah (1960) found that the number of ovarioles in *A. aegypti* varied between 50 and 150. Therefore, the size of the individual female, and the conditions under which the larvae had developed can have a pronounced effect on the number of eggs laid.

Fecundity is also affected by autogeny. In Arctic mosquitoes, many follicles degenerate during autogenous ovarian development and

thus fecundity is so greatly reduced that sometimes only one egg can mature (Corbet, 1964; 1967).

Diapause

Diapause is the most highly evolved system of dormancy for overcoming cyclic, long-term, and extreme environmental conditions. The dormancy is induced well before the adversity and maintained for some time irrespective of environment. It may intervene at any of the major developmental stages of the life cycle, which is always characteristically fixed in each species (Mansingh, 1971).

All species of the genus *Aedes* are capable of arresting development at the egg stage. Diapause induction experiments measure only the results of a sequence of physiological responses which ultimately determine the response of the individual. Each individual has a different stimulus for eliciting this all or none response (DeWilde, 1962). Reactivation from diapause often requires months of extended exposure to environmental conditions such as low temperature, whereas "conditioning" may require a brief exposure to factors such as high humidity (Horsfall, 1956; Harwood and Horsfall, 1959; Clements, 1963).

In *Aedes* eggs, there are two types of diapause, namely obligatory and facultative. The multivoltine species (many generations per year) which are controlled by the environment exhibit facultative diapause. The univoltine species, which are free from environmental control exhibit an obligatory diapause. In Lepidoptera, geographical races of a species often exhibit obligatory and facultative diapause (Mansingh and Smallman, 1967). From the physiological and ecological viewpoint there is no difference between the diapausing generation of multivoltine and univoltine

species (Mansingh, 1971).

The principal stimulus for the onset of diapause is photoperiod although temperature, water, and diet may be involved. DeWilde (1962) reported that photoperiod provided the most reliable indication of seasonal changes although Lees (1956) suggested that both temperature and photoperiod provide token environmental messages, affecting the same endocrine mechanisms.

Temperature plays a major part in termination of diapause. High temperature removes the effect of short photoperiod, low temperature reduces hatching in response to long photoperiod, and prolonged exposure to low temperature terminates diapause (Lees, 1955; Beckel, 1958; de Wilde, 1962; Danilevskii, 1965; McHaffey and Harwood, 1970; Mansingh, 1971; and McHaffey, 1972).

As far as is known *A. campestris* is generally univoltine (McLintock, 1944; Rempel, 1950). However, Chapman (1966) observed that 95% of the eggs obtained from some females of the spring brood hatched after a week of "conditioning". Both Chapman (1966) and Rempel (1950) observed a second brood in the field.

Studies on embryonic diapause in *A. campestris* have not been reported to date, but studies on diapause in *A. dorsalis* have been reported by Khelevin (1958, 1959), Telford (1963), McHaffey and Harwood (1970), and Taylor (1973). Both photoperiod and temperature are important factors in regulating embryonic diapause in *A. dorsalis*.

METHODS AND MATERIALS

General Methods

Collection of Materials

The materials used for laboratory studies were collected from different locations in Manitoba and Saskatchewan. The preserved specimens of *A. melanimon* were obtained from the Canadian National Collection in Ottawa, the United States National Museum, and from the personal collections of J. McLintock (Saskatchewan), R. M. Bohart (California), T. Miura (California), and H. C. Chapman (Nevada).

The different stages of *A. campestris* and *A. dorsalis* were collected from different locations and used for laboratory colonization. Adult females were collected in early summer from areas in Manitoba and Saskatchewan that were known to have fair populations of larvae in pools or a high number of adults per mosquito trap. Adults of *A. campestris* and *A. dorsalis* were collected at Dundurn, Saskatchewan on June 25, 26, 1973; at Estevan, Saskatchewan on June 12, 1974; at Saskatoon, Saskatchewan on June 19-27, 1974; and at Churchill, Manitoba on July 15-17, 1974. The mosquitoes attempting to feed on blood were captured with an aspirator and transferred to the 25- x 25- x 150-mm, acrylic holding cages. The cages had fine-mesh nylon screen on 2 sides to allow the imagos to take blood and to oviposit through the screen. Caged female imagos were transported to the laboratory in insulated chests maintained at 10-15°C, 70-85% R.H. using ice and moist towelling respectively.

Larvae were collected from their developmental sites by dipping and transferring to styrofoam food containers (300 ml) for transport to

the laboratory in insulated chests maintained at 10-15°C.

Larvae and pupae were collected from temporary pools and roadside ditches near Winnipeg on Highway 1 East near the junction with Highway 207. Second-instar larvae of *A. campestris* were found on April 18, 1973, and fourth-instar larvae and pupae were found on May 19, 1975.

Eggs were obtained from the field-collected adults and stored on moist nylon cloth in covered Petri dishes. The technique of keeping eggs moist and free of mold and fungi was similar to the technique used by Kalpage (1970).

Egg storage and hatching

Eggs were kept moist at 23 $\pm$ 1°C and 16L:8D (L:D - Light:Dark). Eggs were kept under these conditions for a minimum of 15 days before they were subjected to the hatching medium. Eggs could be stored successfully for about 9 months.

The eggs were hatched in a nutrient-broth solution (2:1,000 w/w, powder in tap water) maintained at 23°C and 16L:8D.

Determination of diapause

Eggs that failed to hatch after 24 hours in the nutrient-broth solution were bleached and examined to determine viability. It was determined that a period of 24 hours was sufficient to cause non-diapause eggs to hatch. Little or no hatch occurred after this period. The bleaching method developed by Trpis (1970) was found to be very suitable for clearing the egg without destroying the embryo. The viable eggs in a state of diapause were easily distinguished from the dead embryos by the presence of eye spots, hatching spines, and abdominal segmentation.

In some experiments, the eggs were subjected twice to the hatching stimulus, the second one being 30 days after the first. Instead of bleaching, some unhatched eggs in the state of diapause were stored at 5°C for 3 months to terminate diapause. The eggs were again subjected to a hatching stimulus by placing them in a nutrient-broth solution.

Description of the "mating cage"

Mating of the laboratory colony of *A. campestris* occurred within a controlled-environment chamber 120-x 120-x 210-cm high. The chamber which was constructed of 6-mil clear plastic on the inside, and 6-mil black plastic on the outside, was maintained at 70-80% R.H., 23 ± 1°C, and a light:dark condition of 16L:8D.

The light source in the mating cage consisted of four, 25-W., incandescent bulbs arranged in a square (50-cm to a side) above the ceiling. Sixty minutes each of dawn and dusk periods were provided by a 60-W., incandescent bulb at the centre of the square. Each period of 60 minutes occurred prior to and directly after the lighted part of the daily photoperiod cycle. Light was decreased and increased automatically by means of a rheostat driven by a reversing motor.

One side of the mating cage was covered with canvas, and a curved slit fitted with a zipper served as the access or "door". The outside of the door was covered with a black plastic sheet which acted as a curtain to prevent the entrance of external light and the escape of adults when the door was opened.

Carbohydrate and water were provided for adults in the mating cage. Rehydrated apple wrapped in cheese cloth, and cotton balls soaked in water were hung at the corners of the chamber. To prevent desiccation, the cotton balls were moistened periodically.

Determination of mating success in the laboratory

The presence of sperm in the spermathecae was used to determine the percentage insemination or mating success. Ten to 14 days after emergence, a sample of females was aspirated from the "mating cage" (120-x 120-x 210-cm) and dissected on clean slides. The female imagos were anaesthetized with ethyl ether, wetted in 70% ethanol and soaked in saline before dissection. Using 2 dissecting micropins, the spermathecae were removed and examined under a cover slip with a phase-contrast microscope.

Description of photoperiod boxes

All experiments on the effect of photoperiod on the induction of egg diapause were conducted in photoperiod-controlled boxes 22 1/2-x 29-x 24-cm high. The boxes were constructed of wood and fitted with a metal lid. The light source was a 7-V.,0.41-Amp. miniature lamp with the intensity of 39 foot-candles. The light was fixed on top and wired to a timer. The temperature in each light box,which was continually recorded on a thermograph, was maintained at 23 $\pm$ 1°C.

The experiment on termination of egg diapause at 30° C was conducted in a BOD incubator which was equipped with an incandescent light source (40-W. bulb) and a time clock for photoperiod control, 16L:8D. The temperature varied $\pm$ 1°C from the desired setting.

Colonization Techniques

All experiments were conducted under the constant temperature of 23 $\pm$ 1°C, 75 $\pm$ 5% R.H. and 16L:8D. The handling and rearing procedures used for the various developmental stages of *A. campestris* were as follows:

Eggs - These were obtained from field-collected females which had oviposited in the laboratory. The eggs were held for a minimum period of two weeks at 23°C, 16L:8D, before they were hatched. The eggs that failed to hatch in the nutrient broth medium within 24 hours were bleached and examined under the microscope to determine viability.

Larvae - The newly hatched larvae were transferred from nutrient broth solution into covered plastic pans (6-x 22-x 30-cm) containing ca. 1000 ml. of dechlorinated tap water. About 150 to 200 larvae were reared in the same pan. The film of dust or scum on the surface of the water in each pan was removed by daily towelling (see Taylor, 1973). This technique prevented fouling of the larval media and greatly increased the percentage survival of the larvae. In the early phases of this study, larvae were fed with 70-150 mg of Tetramin Tube Food 66®. This was later discontinued, and a suspension of liver powder (Nutritional Biochemicals) was used instead. The latter proved to be an adequate diet and was more readily available from local suppliers.

The experiments on temperature effects on larval development were conducted in BOD incubators. All stages of the colony were maintained under these conditions.

Two complete populations of *A. campestris* were reared at 15, 20, 23, 25, and 27° C. The hours spent in each stage were calculated from the time 50% of one stage reached the next stage. This technique for counting and calculating followed the method used by Brust (1967).

Pupae - Pupae were removed from the larval pans daily and transferred to styrofoam food containers (50 pupae per 30 ml container) half-filled with dechlorinated water. The pupal containers were placed in an acrylic cube cage 30-x 30-x 30-cm. Rehydrated apple and moist paper wicks provided carbohydrate and water for the adults. The

relative humidity inside the cage was maintained at 70-80% by moist paper wicks. Experiments similar to those described for larvae were also conducted with pupae.

For individual rearing, the pupa was transferred to a clean vial 23-x 85-mm, three-quarters-filled with dechlorinated water. A small screen cup was placed over the vial to contain the imago. The screen cup was made of a 15 mm length of acrylic tubing (25-mm diam.) covered with a fine-mesh nylon screen.

Adults - The emerging adults were aspirated daily from the acrylic cube cages and released into the mating cage. After 2 weeks of confinement in the mating cage, the adults were removed. The female imagos were placed in small 25-x 25-x 150-mm, acrylic holding cages (25 females per cage) and were offered a blood meal. Paper towelling moistened with tap water was used as the oviposition substrate. Adult females were fed on an anaesthetized mouse, by placing the mouse on top of the screened portion of the cage on alternate days. Rehydrated apple was placed on the screen to serve as a carbohydrate source for the imagos.

In experiments using eggs from individual female imagos, a circular pad of paper towelling (30-mm diam.) was placed under a small cylindrical cage (15-x 25-mm diam.) which held the female. The eggs were collected after the death of adults and placed on moist nylon pads in Petri dishes and stored.

Introduction, maintenance, and termination of egg diapause

The experiments on induction, maintenance, and termination of egg diapause were carried out using photoperiod as the experimental variable. Only 2 temperatures were employed to determine the effects on diapause.

The field-collected females were allowed to lay their eggs through the screen of 25- x 25-x 150-mm,acrylic holding cages in the laboratory at 23 $\pm$ 1°C, 16L:8D. The eggs were collected and stored on moist nylon cloth in covered Petri dishes in the laboratory at 23 $\pm$ 1°C, 16L:8D for a minimum of 15 days before they were subjected to the different photoperiods at 23°C or 30°C.

The results of the egg-hatching experiments revealed that the Saskatoon populations were primarily multivoltine (99%) whereas Churchill populations were partly univoltine. In order to investigate the effect of a stimulus on the induction of diapause in eggs, the eggs had to be non-diapausing ones. Therefore, the Saskatoon population was used for the investigation on diapause induction and maintenance, while the diapausing eggs from Churchill populations were used to test diapause termination.

Preliminary experiments revealed that there was a high incidence of diapause in the eggs stored at a short photoperiod, 12L:12D, and even more at 8L:16D while the eggs stored at a long photoperiod (16L:8D) exhibited little or no diapause effect.

Experiments were conducted at 23 $\pm$ 1°C and photoperiods of 8L: 16D, 12L:12D, 13L:11D, 14L:10D, 15L:9D, and 16L:8D. The eggs were stored at a given photoperiod for 4, 7, 10, 14, 20, 30, and 45 days respectively. At the end of each trial period, the eggs were immersed in nutrient broth solution.

The non-diapause eggs from Churchill populations were allowed to hatch in nutrient broth solution. The unhatched eggs which were in a state of diapause were then used in the diapause-termination experiment.

The diapausing eggs were stored for periods of 4, 7, 10, 14, 20, and 30 days at two temperatures, 23 $\pm$ 1°C and 30 $\pm$ 1°C, and a photoperiod of 16L:8D. At the end of the trial period, the eggs were immersed in a nutrient broth solution.

RESULTS

Determination of voltinism in different populations of *A. campestris*

Eggs from field-collected females were stored at 16L:8D, 23 ± 1°C and hatched. Eggs which did not hatch the first time were subjected to a second hatching stimulus 30 days after the first one. The eggs that failed to hatch the second time were bleached and examined under a dissecting microscope. The results in Table 1 indicate that the populations from Estevan, Saskatoon, Winnipeg, and Churchill are primarily multivoltine. All eggs of Winnipeg populations hatched during the first flooding. Only a portion of the eggs from the Churchill population hatched under identical conditions. After two floodings, I determined that 35.88% of the Churchill egg populations were in diapause. After two floodings I determined that less than 1% of the Estevan and Saskatoon egg populations were in diapause (Table 1).

Effect of temperature on larval, pupal, and adult development

At a photoperiod of 16L:8D, five temperatures (15, 20, 23, 25, and 27°C) were used to investigate the effect of temperature on the development of *A. campestris* from Saskatoon and Churchill.

The results indicate that time of development for all immature stages was dependent upon temperature. The larval and pupal development was more rapid at the highest temperature (27°C) than at any of the others. Survival of the larvae was greatly reduced at 15°C. Survival was greatest at 23°C (Table 2 and Fig. I). No record was made on the weight and size of the immatures but larvae, pupae, and

Table 1. Determination of voltinism in different populations of *A. campestris*.

TOTAL NO. EGGS	NO. EGGS HATCHED 1st HATCH	NO. EGGS HATCHED 2nd HATCH	NO. EGGS UNHATCHED	NO. VIABLE EGGS	% UNIVOLTINE (DIAPAUSE EGGS)	% MULTIVOLTINE
		Estevan				
1217	774	136	5	915	0.55	99.5
		Saskatoon				
400	262	2	4	268		
322	247	0	0	247		
271	195	2	0	97		
304	286	0	3	289		
157	112	5	1	118	0.7	99.3
		Churchill				
1132	427	0	419	846		
1500	563	31	174	768		
750	303	0	129	432		
650	229	0	82	311		
950	329	0	249	578	35.88	64.12
		Winnipeg				
200	117	0	0	117		
200	120	0	0	120		
200	108	0	0	108		
200	124	0	0	124		
200	109	0	0	109	0	100

Table 2. Effect of temperature on development and survival of *A. campestris*.

DEVELOPMENT STAGE	TEMPERATURE (°C)	CHURCHILL POPULATION[1]		SASKATOON POPULATION[1]	
		Time (hours)[2] Mean ± s.d.[5]	% Mortality Mean ± s.d.	Time (hours)[2] Mean ± s.d.	% mortality Mean ± s.d.
Larval Instar I	15	0 + 24	0	0 + 24	0
	20	0 + 24	0	0 + 24	0
	23	0 + 24	0	0 + 24	0
	25	0 + 24	0	0 + 24	0
	27	0 + 24	0	0 + 24	0
New II	15	122 ± 3.5	11 ± 3.5	139 ± 4.2	8.5 ± 5.4
	20	54 ± 3	2 ± 0.7	66.5 ± 3.5	20 ± 5.1
	23	46 ± 1.4	3 ± 2.2	48 ± 1.5	5 ± 2.1
	25	36.5 ± 0.7	2 ± 0	46 ± 1.4	3 ± 2.2
	27	34 ± 1.4	1 ± 0.7	42 ± 2.8	14 ± 3.0
New III	15	220 ± 8.1	97 ± 2.5	289 ± 11	38 ± 9.8
	20	121 ± 5.0	19 ± 8.4	137 ± 3.2	35 ± 5.6
	23	86 ± 1.5	9 ± 5.2	98 ± 2.4	10 ± 3.0
	25	72.5 ± 1.4	13 ± 6.5	96 ± 2.8	16 ± 4.8
	27	71 ± 2	26 ± 7.2	94 ± 1.5	21 ± 6.8
New IV	15	384 ± 8.1	100	524 ± 121	44 ± 7.1
	20	193 ± 7	55 ± 9.1	238 ± 8.4	58 ± 10.7
	23	150 ± 8.8	17 ± 6.3	162 ± 8.3	24 ± 7.1
	25	142 ± 5	41 ± 4.2	158 ± 10.1	46 ± 5.1
	27	140 ± 6.1	50 ± 6.1	139 ± 7.1	45 ± 3.5

Table 2. cont.

mature IV (pharate pupa)	15	-	-	696 ± 5.65	98 ± 2.8
	20	317 ± 9.9a[3]	70 ± 11.3a	360 ± 11.31a	66 ± 7.1a
	23	210 ± 4.2b	23.5 ± 4.9b	222 ± 8.48 b	28 ± 4.2b
	25	192 ± 5.6b	41 ± 8.5bc	211 ± 4.2b	49 ± 5.7a
	27	190 ± 4.2b	55 ± 7.1ac	200 ± 2.8b	60 ± 7.1a
mature pupa (pharate adult)	15	-	-	858	100
	20	S[4] 410 ± 7.07a	75 ± 7.1a	S 452±7.1a	68 ± 9.9a
	23	285 ± 5.7b	30 ± 5.2b	294 ± 4.2b	40 ± 5.6b
	25	256 ± 5.6c	42 ± 2.8b	280 ± 5.6b	59± 5.6a
	27	S 243 ± 4.2c	65 ± 3.5a	S 260 ± 2.8c	70 ± 4.2a

1. Mean of 2 replicates

2. Time for 50% of one instar or stage to molt to the next instar or stage.

3. Common letter following column indicates no significant difference at the 5% level between pupal and larval instar IV development rates and mortality at different temperatures. (Student Newman Kewl test).

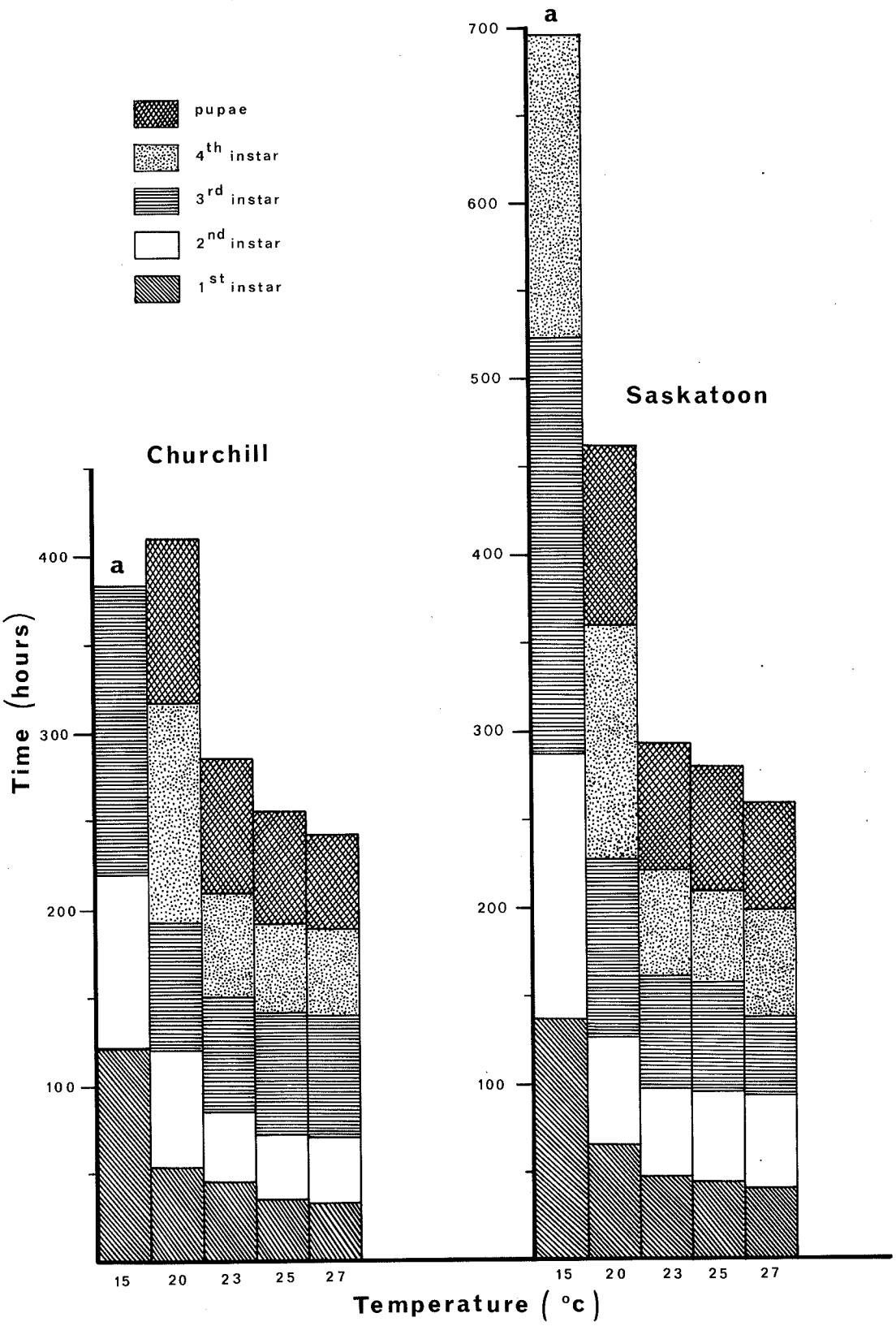
4. Letter "S" preceding column indicates a significant difference at the 5% level in pupal and larval instar IV development rates and mortality between Churchill and Saskatoon populations. F-test for analysis-of-variance.

5. Abbreviation: s.d., standard deviation.

Figure 1.

Rate of growth of *A. campestris* larvae at 5 temperatures.

a) Larvae did not survive to the next stage of development.


pupae
4th instar
3rd instar
2nd instar
1st instar
Churchill
a
Saskatoon
a
Time (hours)
100
200
300
400
500
600
700
15
20
23
25
27
Temperature (°c)

adults reared at 23°C appeared to be larger than those reared at the other temperatures.

The results in Table 2 indicate that there was no significant difference in the development time and the % mortality between the Saskatoon and the Churchill populations at all temperatures used, with 2 exceptions. The development time for pupae was greater in the Saskatoon population at 20°C and 27°C.

Since the development time shown for the mature instar IV larva is a cumulative figure derived from adding the time spent in the previous larval instars, a statistical analysis is shown for this final larval stage only. Any analysis of the data given for the preceding larval instars would be redundant.

Maturation of ovarian follicles

In this study, female imagos of *A. campestris* from Churchill and Saskatoon were examined in order to determine their normal mode of follicular maturation.

Female imagos, aged 5 to 7 days, were blood fed. The females were then anaesthetized with ether, wetted in 70% ethanol and soaked in a saline solution before being dissected. The dissection was carried out on a slide using a few drops of fresh saline. The intact ovaries were removed using the technique of Christophers (1960), and transferred to another slide with a drop of saline, and then covered with a glass cover slip. The ovaries were immediately examined under a phase-contrast microscope.

Follicular maturation was graded according to the following classification scheme, adapted from Christophers (1911) and Clements (1963).

Stage 1 a: follicle is more or less spheroid and consists of a small group of comparatively small, not easily differentiated cells. No differentiation of oocyte from nurse cells (Fig. 2a).

1 b: follicle is spherical, contains a differentiated oocyte nucleus and 7 nurse cells surrounded by a distinct follicular epithelium (Fig. 2b).

Stage 2 a: follicle is spheroid and larger. A few very fine yolk granules appear around the oocyte nucleus but they do not obscure the oocyte nucleus (Fig. 2c).

2 b: follicle is slightly ovoid, a few coarse yolk granules appear around the oocyte nucleus until the nucleus is quite hidden; the follicle is in the resting stage (Fig. 2d).

Stage 3 a: follicle is slightly ovoid, oocyte cytoplasm is cloudy with yolk granules and no longer clearly distinguishable in the fresh preparation. About half to two-thirds of the follicle is filled with yolk (Fig. 2e).

3 b: the yolk obscures the oocyte nucleus, and about two-thirds to three-quarters of the follicle is filled with yolk (Fig. 2f).

Stage 4 a: follicle is ovoid, starts to elongate, and yolk occupies nine-tenths of the follicle (Fig. 2g).

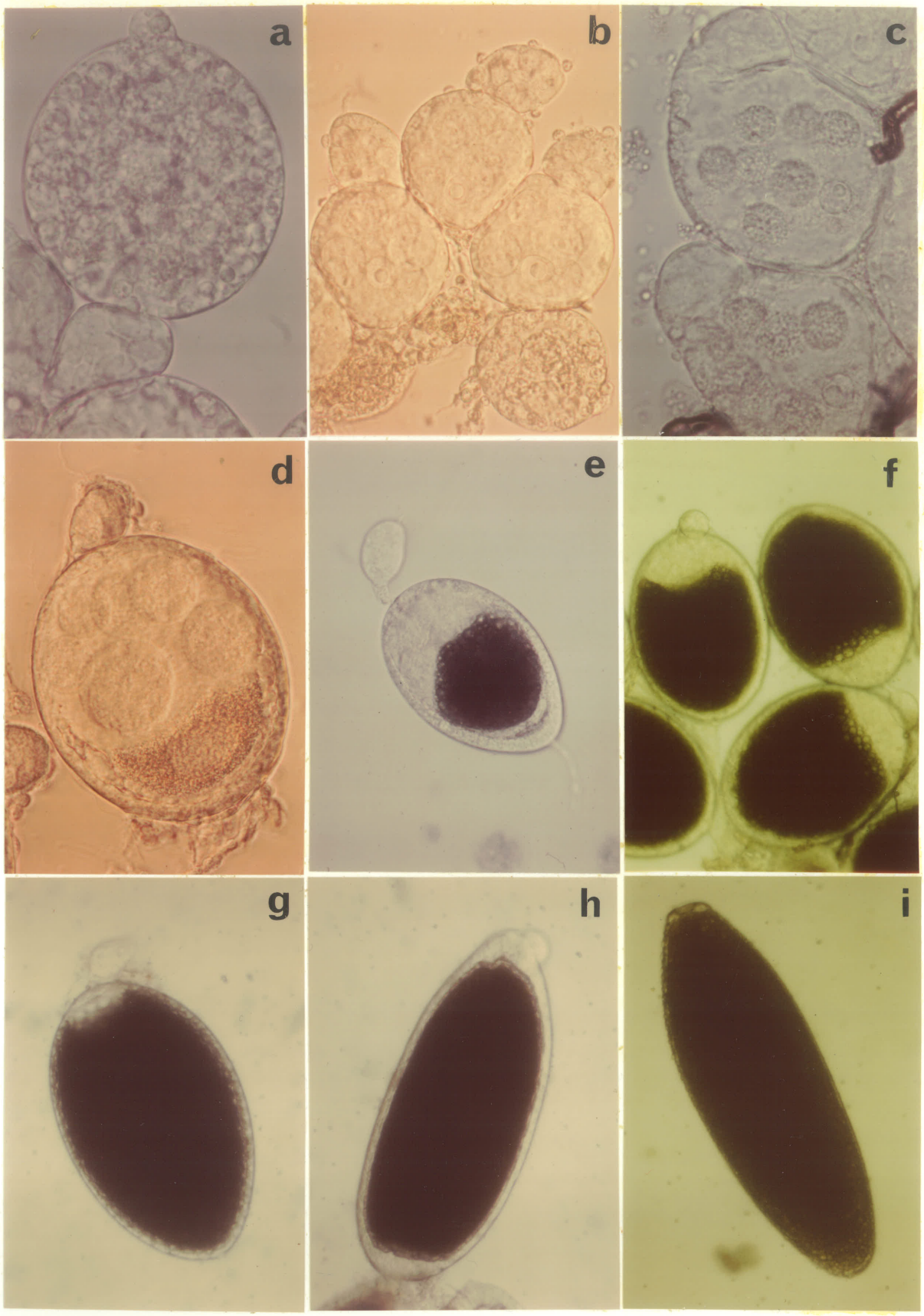
4 b: follicle is elongate and assumes the stage of the mature egg (Fig. 2h).

Stage 5: chorionic structure becomes visible. Egg is mature and ready for laying (Fig. 2i).

In *A. campestris*, the stage of follicular development of the

Figure 2 a-i

Stages of follicular maturation in female imagos of *A. campestris:* (a) stage 1 a (160x); (b) stage 1 b (100x); (c) stage 2 a (125x); (d) stage 2 b (100x); (e) stage 3 a (25x); (f) stage 3 b (25x); (g) stage 4 a (40x); (h) stage 4 b (40x); (i) stage 5 (40x).


a
b
c
d
e
f
g
h
i

ovaries at emergence and shortly thereafter ranges from 1 to 2a.

The results in Table 3 indicate the rate of follicular development from the resting stage to maturity in blood-fed female imagos of *A. campestris* at 23°C. The female imagos could complete their gonotrophic cycle within 3-4 days after taking a blood meal (Fig. 3 a-c). No significant difference occurred between the rate of follicular development in females from Churchill and females from Saskatoon (Table 3).

Autogeny in *A. campestris*

In this experiment, all Churchill and Saskatoon populations were reared at 23 ± 1°C, 75% RH, and 16L:8D. The emerging adults of each population were maintained in similar cages (30-x 30-x 30-cm) and provided with water and a carbohydrate source. The non-blood-fed females were sampled at various times (days) following emergence and dissected, and the stage of follicular development was recorded. Females with some of their follicles matured beyond stage 2 b, to 3 a, were recorded as being facultatively autogenous. The results as shown in Table 4 indicate that facultative autogeny occurred in populations from Churchill and Saskatoon. A greater percentage of autogeny occurred in the Churchill populations, where 45% of the females demonstrated autogenous development versus 13.9% for the Saskatoon population (Table 4). Fig. 4 shows the facultatively autogenous eggs in *A. campestris* from Saskatoon. Only some follicles developed beyond the resting stage in both the Saskatoon and the Churchill females. Mature eggs were not numerous, and in most cases very few follicles reached maturity. This, plus the fact that development was delayed until the females were 15-20 days old are the reasons for using the term, "facultative," to describe autogenous development in *A. campestris* (See Ellis and Brust, 1973).

Table 3. Rate of follicular development in blood-fed females of *A. campestris* at 23°C.

TIME AFTER BLOOD MEAL (days)	NO. FEMALES DISSECTED	STAGE OF FOLLICULAR DEVELOPMENT					
		2b	3a	3b	4a	4b	5
		Churchill Population					
1	7	4	2	1	-	-	-
2	5	-	1	3	1	-	-
3	7	-	-	-	1	4	2
4	8	-	-	-	1	1	6
5	5	-	-	-	-	-	5
		Saskatoon Population					
1	5	1	3	1	-	-	-
2	5	-	1	3	1	-	-
3	5	-	-	1	1	2	1
4	5	-	-	-	-	-	5
5	5	-	-	-	-	-	5

Figure 3 a-c

Stages of follicular maturation at 1, 2, and 3 days after a blood meal in female imagos of *A. campestris* reared at 23°C :

(a) 1 day after a blood meal (40x);

(b) 2 days after a blood meal (40x);

(c) 3 days after a blood meal (25x).

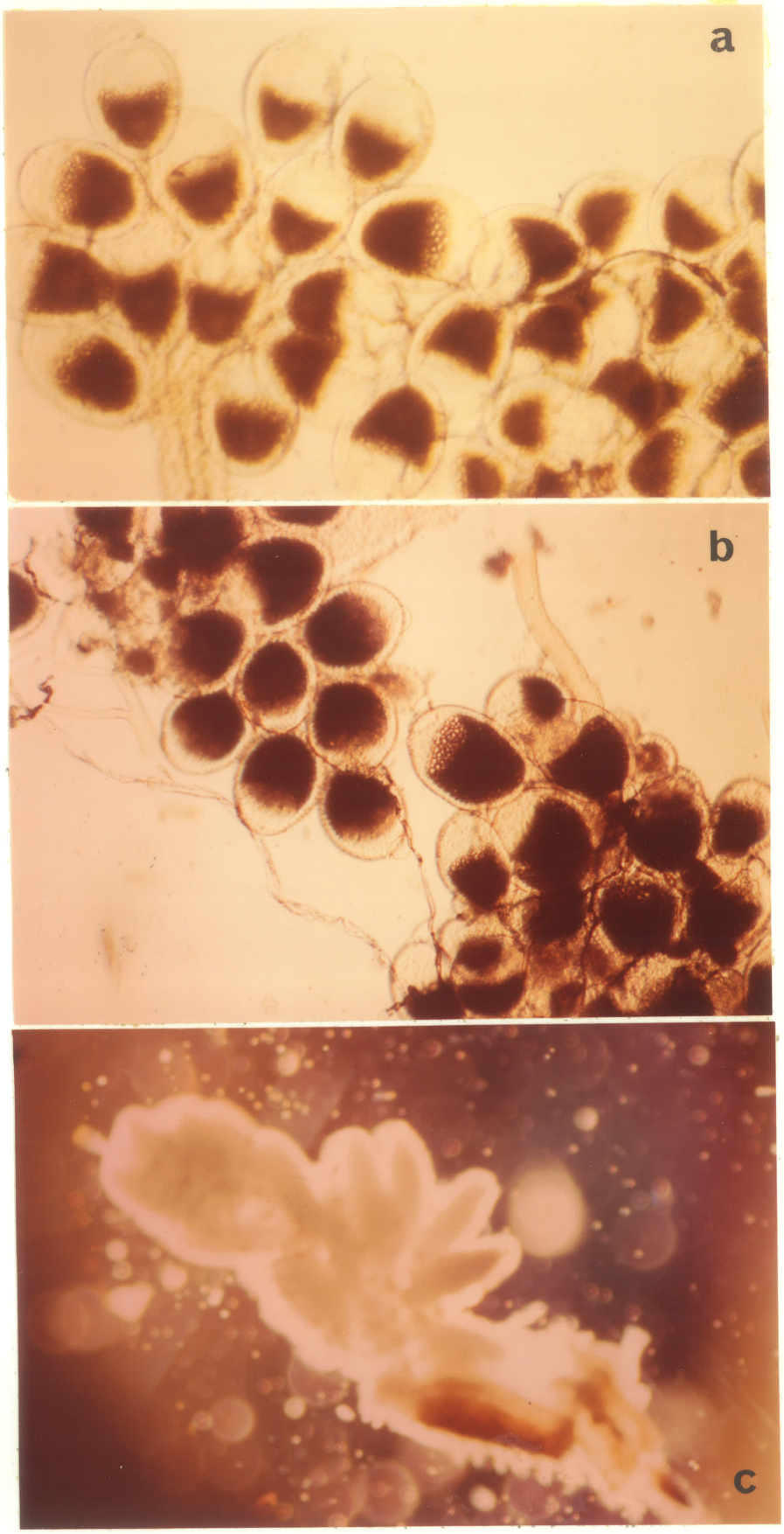

a
b
c

Table 4. Autogenous egg development in *A. campestris*, from Churchill and Saskatoon, reared at 23°C and photoperiod 16L:8D.

AGE IN DAYS AT DISSECTION	NO. FEMALES DISSECTED	STAGE OF OVARIAN FOLLICLES									% AUTOGENY
		1a	1b	2a	2b	3a[1]	3b	4a	4b	5	
					Churchill population						
10-12	10	-	2	-	7	1	-	-	-	-	
15-20	15	-	-	2	7	1	-	1	-	4	(78)[2]
25-30	15	-	-	1	3	8	-	2	-	1	45%
					Saskatoon population						
10-12	8	-	2	4	2	-	-	-	-	-	
15-20	15	-	1	4	7	-	1	-	-	2	(22)[2]
25-30	13	-	-	5	6	2	-	-	-	-	13.9%

1. Follicles developed beyond stage 2b to 3a are considered as autogenous eggs.

2. Figures in parenthesis indicate number of eggs laid by autogenous females.

Figure 4

A group of follicles of a facultatively autogenous female of *A. campestris* from Saskatoon, Saskatchewan (23 days old; 25x).

Mating success in *A. campestris*

In this study, all experiments were conducted in the same cage under similar conditions (as described for the mating cage) with populations of the F_1 generation. The percentage insemination of the female imagos was used to determine mating success. Very little mating occurred before 7 days in the cage, and females were not removed until 14 days. A subcolony was established through 3 generations in the populations from Churchill and Saskatoon. Only 1 generation was reared from the Estevan population, due to inadequate specimens to maintain a laboratory colony.

The results shown in Table 5 indicate that there was no increase in the percentage of cage mating, and no increase in the % egg viability in *A. campestris* within 3 generations in the Saskatoon population. Due to the variation in the number of adults per generation in the Churchill population, it is difficult to assess the effect of multiple generations on the incidence of cage mating and on egg viability.

Effect of the number of blood meals on female fecundity

The unmated females from Saskatoon (5-7 days old) were given 1, 2, or 3 blood meals, with an interval of 2 days between each feeding. The females were then allowed to lay eggs individually under a small screen cup (1.5-cm high x 2.5-cm diam.). The eggs were collected and counted after the female died.

A second set of experiments was conducted the same way using mated females. A population from Estevan was used, and females 5-7 days old were force-mated (McDaniel and Horsfall, 1957) and blood-fed. Females from Churchill were also used in this experiment, but these were taken from the mating cage and blood-fed after they were 14 days old.

The results in Table 6 indicate that there is no significant

Table 5. Mating success of *A. campestris* in 120-x 120-x 210-cm cage.

SOURCE	GENERATION	TOTAL NO. ADULTS	CONFINEMENT PERIOD(DAYS)	% INSEMINATION[a]	% EGG VIABILITY[b]
Churchill	1	2594	16	22.2	19.7
	2	1747	15	30	35
	3	873	16	20	18.3
Saskatoon	1	1218	14	25	21
	2	3657	15	22.5	17
	3	1272	15	20	20
Estevan	1	774	15	25	24.9

a) Number of females ranged from 10-18.

b) Mean of 3 replicates.

Table 6. Effect of the number of blood meals on the fecundity of *A. campestris* at 23°C.

SOURCE	NO. OF BLOOD MEALS	NO. OF FEMALES	TOTAL EGGS LAID	NO. EGGS LAID PER FEMALE		
				MIN.	MAX.	MEAN ± S.E.[1]
			Mated females			
Estevan	1	34	3684	3	243	108.35 ± 9.31a[2]
	2	36	3769	6	190	104.69 ± 8.87a
	3	44	5549	13	212	126.11 ± 7.27a
Churchill	1	20	702	2	115	35.1 ± 8.30a
	2	42	2911	1	163	69.31 ± 7.18b
	3	79	5138	1	186	65.04 ± 5.73b
			Unmated females			
Saskatoon	1	15	1472	24	161	98.13 ± 12.17a
	2	15	1362	3	190	90.8 ± 18.72a
	3	28	2674	2	209	95.5 ± 13.83a

1) S.E. = standard error.

2) Common letter following column indicates no significant difference at 5% level between number of eggs laid by females from each location. Student Newman Kewl test.

difference between the number of eggs produced by females fed 1, 2, or 3 blood meals in populations from Estevan and Saskatoon. The number of eggs produced by Churchill females given 2 or 3 blood meals appears to be greater than by females fed 1 blood meal. Comparisons cannot be made in regard to the fecundity of females from different geographic areas, as the populations were not handled in the same manner.

Effect of photoperiod on the induction of egg diapause

The eggs from the Saskatoon population were used in this experiment. The results as shown in Table 7 and Fig. 5 indicate that the critical period for induction of egg diapause in the Saskatoon populations occurred between 13L:11D and 14L:10D. The greatest percentage of diapausing eggs was obtained after 30 days of exposure to 13L:11D, 12L:12D or 8L:16D. The percentage of diapausing eggs increased sharply after 10-14 days (Fig. 5).

Effect of photoperiod and temperature on the termination of egg diapause

The diapausing eggs from Churchill were used in this experiment. The results as shown in Table 8 indicate that at 16L:8D, 23°C, some eggs began to come out of diapause after 7 days. At 30°C, even more eggs hatched, indicating that diapause had been terminated in these eggs. The greatest percentage egg hatch was obtained at 30°C after 10 to 30 days (Table 8).

Table 7. Effect of photoperiod on the induction of diapause in multivoltine eggs of *A. campestris* from Saskatoon, at 23°C.

NO. DAYS EXPOSURE	PHOTOPERIOD (L:D)	TOTAL NO. VIABLE EGGS	NO. EGGS HATCHED	NO. EGGS UNHATCHED	% DIAPAUSE IN EGGS ± S.E.[1]
4	8:16	288	264	24	8.47±1.32a[2]
	12:12	253	232	21	8.36±0.68a
	13:11	195	189	6	3.04±1.55b
	14:10	301	290	11	3.67±1.51b
	15:9	268	256	12	4.39±1.14b
	16:8	262	254	8	3.03±0.27b
7	8:16	265	240	25	9.49±2.0a
	12:12	276	249	27	9.73±1.71a
	13:11	236	204	32	13.12±4.43a
	14:10	278	269	9	3.06±1.93a
	15:9	296	284	12	4.1±1.62a
	16:8	254	245	9	3.53±1.37a
10	8:16	276	236	40	14.57±1.62a
	12:12	259	234	25	9.64±1.33b
	13:11	249	220	29	11.74±1.04ab
	14:10	258	241	17	6.66±1.75c
	15:9	262	247	15	5.74±0.63c
	16:8	279	270	9	3.23±0.07c
14	8:16	273	185	88	32.03±4.54a
	12:12	282	238	44	15.53±2.35b
	13:11	219	197	22	10.26±2.20b
	14:10	262	256	16	6.13±0.39c
	15:9	259	243	16	6.17±0.15c
	16:8	316	300	16	5.11±0.77c
20	8:16	291	149	142	48.99±3.41a
	12:12	261	175	86	32.91±3.85b
	13:11	207	178	29	14.55±5.18c
	14:10	280	263	17	6.14±1.05d
	15:9	261	251	10	3.82±0.97d
	16:8	318	309	9	2.9±1.07d
30	8:16	288	88	220	73.13±2.60a
	12:12	265	114	151	56.83±1.60b
	13:11	201	150	51	25.23±4.39c
	14:10	288	269	19	6.50±3.19d
	15:9	239	224	15	6.19±1.03d
	16:8	301	297	4	1.18±1.18d

Table 7. cont.

45	8:16	213	73	140	65.80±0.82a
	12:12	199	91	108	53.89±2.75b
	13:11	254	207	47	18.41±3.32c
	14:10	302	281	21	6.96±2.55d
	15.9	237	223	14	5.9±0.31d
	16:8	258	254	4	1.5±1.50d

1. Mean of 3 replicates.

2. Common letter following column indicates no significant difference at 5% level between percentage of diapause in eggs at different photoperiods. Student Newman Kewl test.

S.E. Standard error.

Figure 5

Effect of different photoperiods on the induction of egg diapause in *A. campestris* (Saskatoon population) at 23°C.

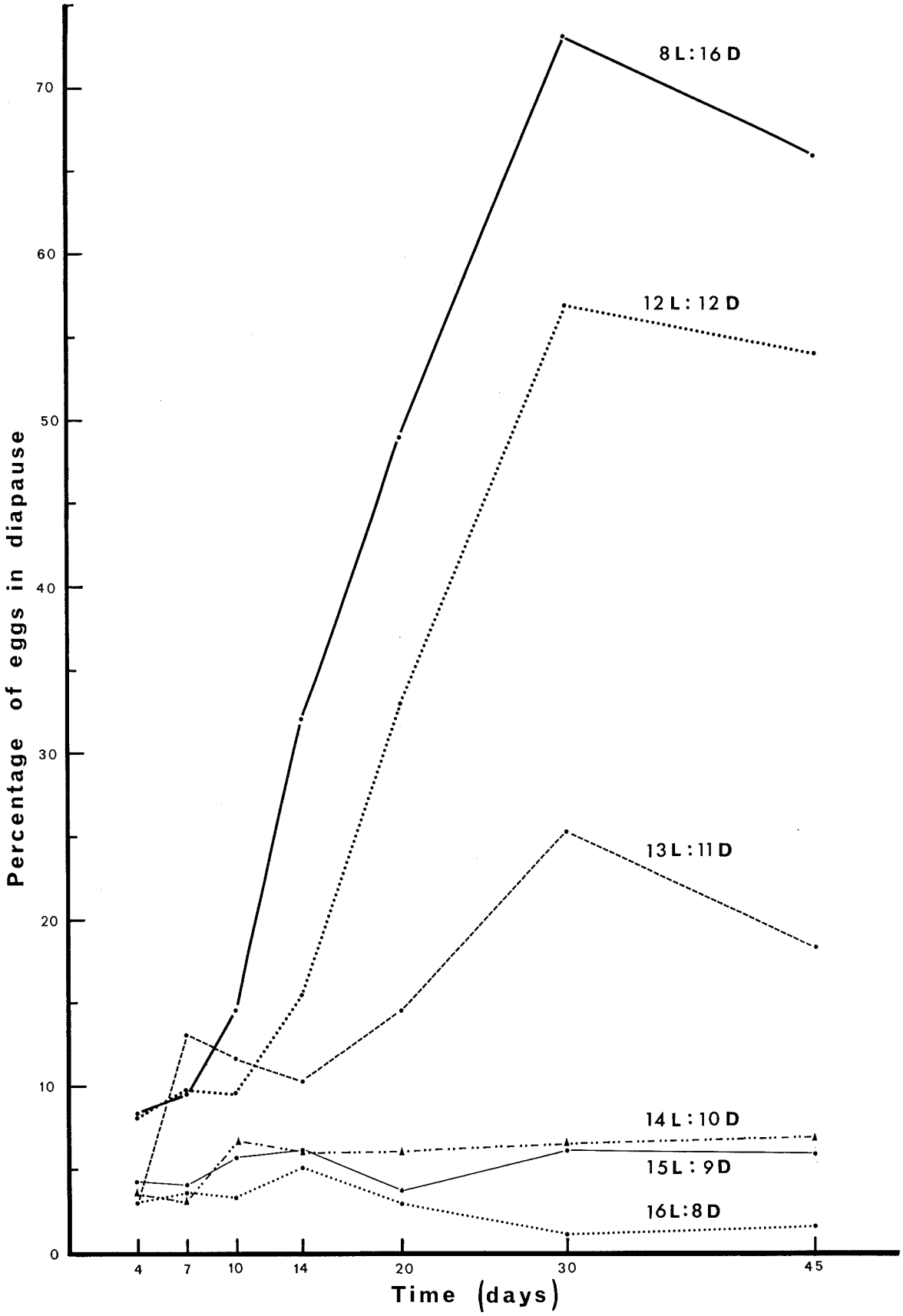

8 L : 16 D
12 L : 12 D
13 L : 11 D
14 L : 10 D
15 L : 9 D
16 L : 8 D
Percentage of eggs in diapause
0
10
20
30
40
50
60
70
4
7
10
14
20
30
45
Time (days)

Table 8. Effect of long photoperiod and temperatures on termination of diapause in eggs of *A. campestris* from Churchill. Eggs maintained at 23°C and 30°C, photoperiod 16L:8D.

NO. DAYS EXPOSURE	TEMP. (°C)	NO. EGGS HATCHED	TOTAL EGGS VIABLE	% HATCH[1] ± S.E.
4	23	3	192	1.56 ± 0.59a[2]
	30	18	322	5.59 ± 1.08a
7	23	14	199	7.03 ± 1.43a
	30	40	238	16.70 ± 1.91b
10	23	38	191	19.39 ± 3.22a
	30	114	193	N[3] 59.07 ± 1.60b
14	23	33	193	17.1 ± 1
	30	-	-	-
20	23	43	172	25 ± 2.16a
	30	140	247	N 56.68 ± 1.47 b
30	23	80	189	42.32 ± 6.95a
	30	91	159	N 57.23 ± 9.40a
45	23	104	215	48.37 ± 1.76
	30	-	-	-

1. Mean of 3 replicates.

2. Common letter following column indicates no significant difference at 5% level between percentage egg hatch at 23 and 30°C. Student Newman Kewl test.

3. Letter "N" preceding column indicates no significant difference at the 5% level between percentage egg hatch after 10 to 30 days of exposure at 30°C. Student Newman Kewl test.

S.E. Standard error.

SPECIES DESCRIPTION

Aedes campestris Dyar and Knab belongs to the genus *Aedes* Meigen, subgenus *Ochlerotatus* Lynch Arribalzaga, of the subfamily Culicinae and the family Culicidae. The subgenus *Ochlerotatus* consists of 8 groups according to adult characters, larval habitat, and geographical areas (Edwards, 1932). *A. campestris* and *A. dorsalis* are grouped into group E (*dorsalis*-group: Acartomyia) according to Edwards (1932). *A. canadensis* was also placed in this group by Edwards (1932). Kalpage and Brust (1968) suggested that *A campestris* and *A. dorsalis* from Manitoba might be grouped separately (I(b) of subgenus *Ochlerotatus)* according to the egg chorionic detail and adult tarsal scale color. They moved *A. canadensis* into group I(a) together with other members of Edwards' Group B.

From the present study I suggest that *A. mediolineata* should be placed in the same group as *A. campestris* and *A. dorsalis*, because of close similarities to these species in both the adult male and female, the egg chorionic detail, and the geographical distribution. This would be Edwards' group E (*dorsalis*-group: Acartomyia) or Kalpage and Brust's group I(b).

The terminology used is based largely on that used by Rempel (1950, 1953), Carpenter and LaCasse (1955), Price (1960), and Knight and Laffoon (1970, 1971).

Aedes (Ochlerotatus) campestris Dyar and Knab.

Aedes campestris Dyar and Knab, 1907, Jour. N.Y. Ent. Soc., 15:213.

Aedes callithotrys Dyar, 1920, Ins. Ins. Mens., 8:16.

FEMALE IMAGO. Medium-to large-sized mosquito, length (from head to the tip of abdomen) 5.3 to 6.5 mm ($\bar{x}$ 5.7).

Head: Proboscis black, intermixed with white and pale scales at the basal half up to two-thirds of the length of proboscis. Palpi short, smaller than one-fourth of the length of proboscis, with black scales and a few white scales intermixed. Antennae dark-brown, with subequal joints, rugose, pilose. Torus yellowish-brown, occasionally dark-brown, with inner surface dark; bears short, broad, white scales on dorsal and inner surface. Clypeus dark-brown, cone-shaped. Vertex with short, narrow, curved, yellowish-white scales; and a series of pale golden setae. Occiput black, covered with narrow, curved, pale yellowish to white scales, and erect, forked, yellowish scales in a broad median zone; a small submedian patch with narrow, curved, brown scales and dark, erect, forked scales with a series of black bristles along the hind edge of eyes; lateral zone with broad yellowish-white scales. Thorax: (a diagram of the parts of the thorax of an *Aedes* mosquito is shown in Fig. 6 A, B). Anterior pronotal lobe elliptical, pilose dorsally; covered with dark-brown bristles at the tip of inner part; narrow, curved, yellowish-white scales below sprinkled with yellowish-white setae. Scutum with dark-brown integument; broad median stripe covered with short, narrow, curved, yellowish-brown or dark-brown scales, posterior edge covered with narrow, curved, white scales; lateral margins dark-brown; remainder of scutum covered anteriorly with narrow, curved, white scales and a patch of yellowish-brown scales at the posterior end (Fig. 7B). Posterior pronotum with narrow, curved, brown scales dorsally; narrow, white scales ventrally. Scutellum trilobed, pale yellow to yellowish-brown integument; with narrow, curved, white

Figure 6

Parts of the thorax and pleural chaetotaxy of an adult *Aedes* mosquito.

(A) Lateral view with sclerites and positions of bristles indicated.

(B) Dorsal view of the Thorax.

1. anterior pronotum.
2. propleuron
3. posterior pronotum
4. postspiracular area
5. prealar area
6. mesepisternum
7. mesepimeron
8. metepisternum
9. metasternum
10. metepimeron
11. mesopostnotum
12. mesomeron
13. Hypostigma

ApS, antepronotal seta. C, coxa. H, halter.

MeSL, lower mesepimeral seta.

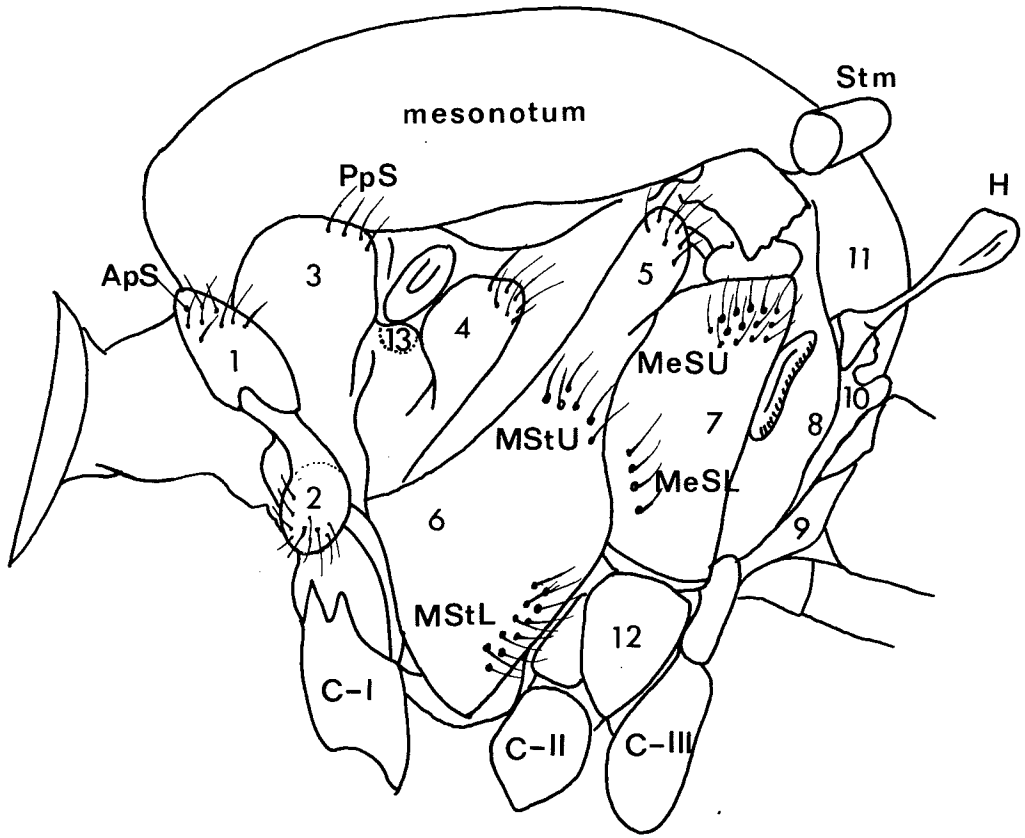
MeSu, upper mesepimeral seta.

MStL, lower mesepisternal seta.

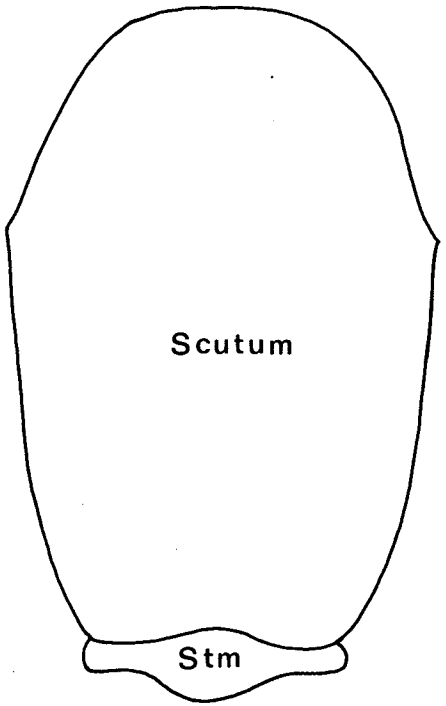
MStU, upper mesepisternal seta.

PpS, postpronotal seta.

Stm, scutellum.


mesonotum
Stm
PpS
H
ApS
3
5
11
1
13
4
MeSU
10
7
8
MStU
MeSL
2
6
9
MStL
12
C-I
C-II
C-III

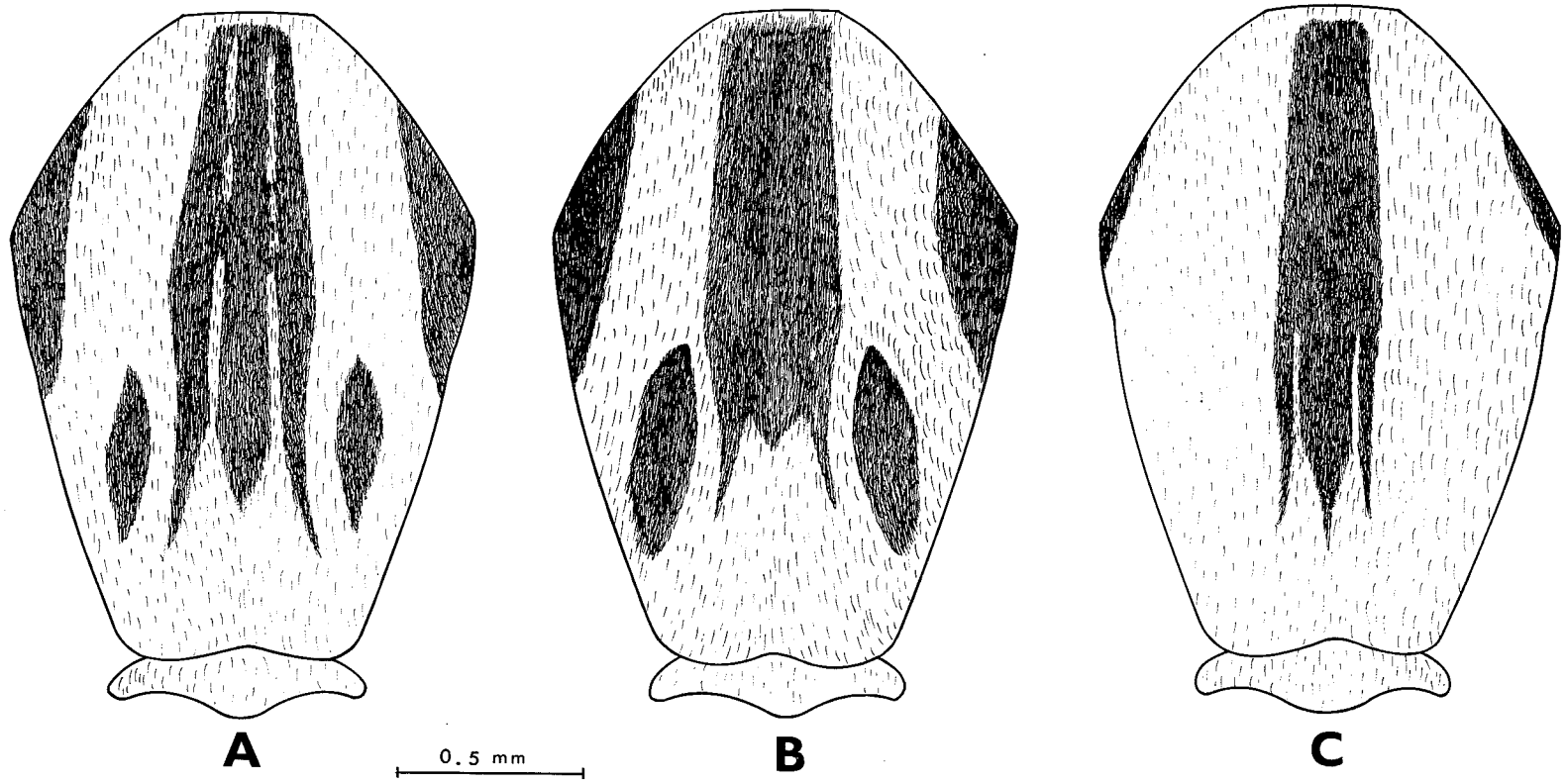
A


Scutum
Stm

B

Figure 7

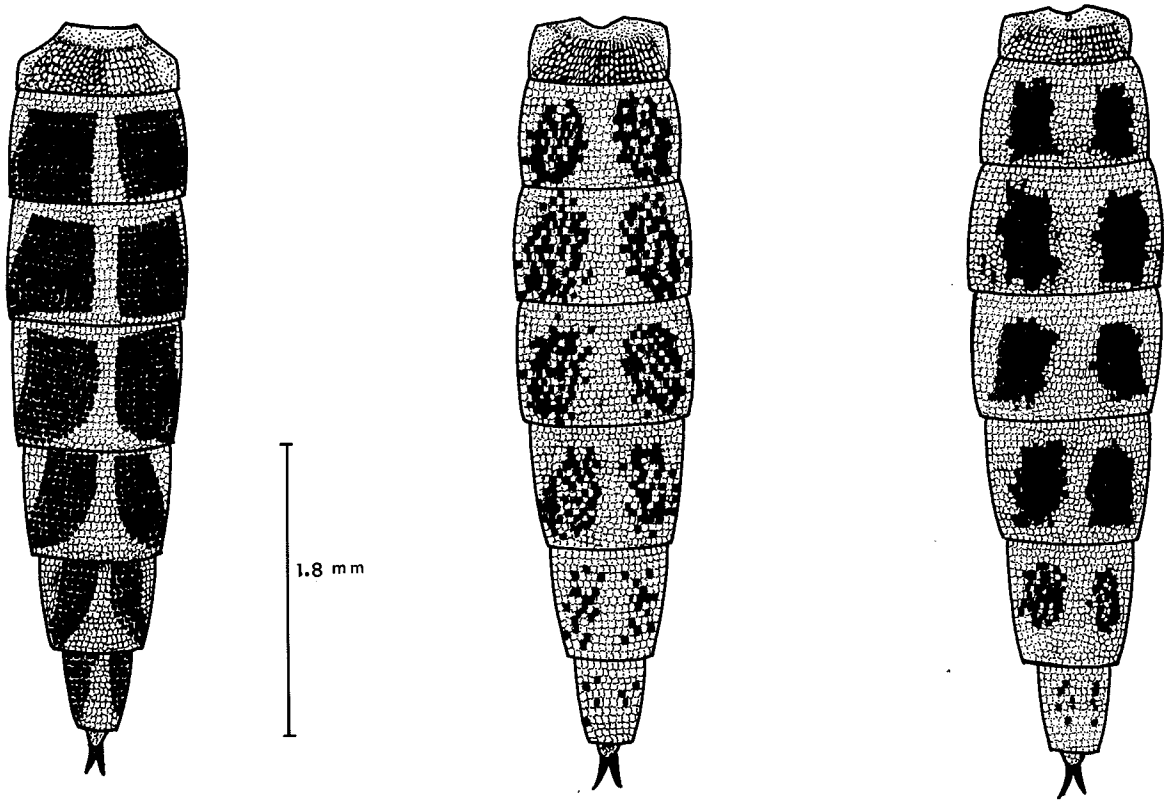
Dorsal view of the thorax of the adult females. (A) *A. mediolineata*. (B) *A. campestris*. (C) *A. dorsalis*.


A
0.5 mm
B
C

scales and pale golden setae along the lobes. Postnotum cup-shaped, prominent, nude, usually yellowish-brown, occasionally yellow. Pleura with extensive poorly defined patches of broad, dull-white scales down to the coxa. Mesepisternal scale patch extending to anterior angle, continuous with prealar scale patch. Mesepimeral scale patch extending to lower margin. Hypostigma with many white scales. Lower mesepimeral bristles about 3-7 ($\bar{x}$ 4.54). Abdomen: Subcylindrical, flattened, posterior segments tapering. Dorsal appearance with sordid-white scales and two patches of dark and white scales intermixed on segments 2-6 (Fig. 8). First tergite with median patch of broad white scales and numerous long white hairs. Venter covered with white scales. Remaining tergites with white scales surrounded by dark, irregular submedian patches on each tergite; each dark patch with sprinkling of a few white scales. Seventh segment almost entirely white except for a few dark scales. Cerci black. Sternites white scaled, with irregular patches of dark scales on median and submedian areas. Legs: slender; Femur: anterior surface mostly white scales with dark scales intermixed; posterior surface creamy white scales. Tibia: white scales with sprinkling of dark scales on anterior surface. Tarsi: First tarsal segments white with dark scales intermixed. Foretarsi dark with narrow, white basal band on segments 2 and 3; segments 4 and 5 dark. Midtarsi, segments 2, 3 dark with narrow, white, basal and apical bands; segment 4 dark with narrow, white basal band; segment 5 almost entirely dark. Hindtarsi dark with narrow white basal and apical bands on segments 2, 3, and 4; segment 5 white, with speckles of dark scales. Tarsal claws with primary tooth gradually curved and 2.1 times as long as secondary tooth (Fig. 9, Fig. 10: B_1, B_2). Claw ratio (Length of primary tooth in relation to the length of basal portion of claw) 1.49:0.98. Wings: White scaled

Figure 8

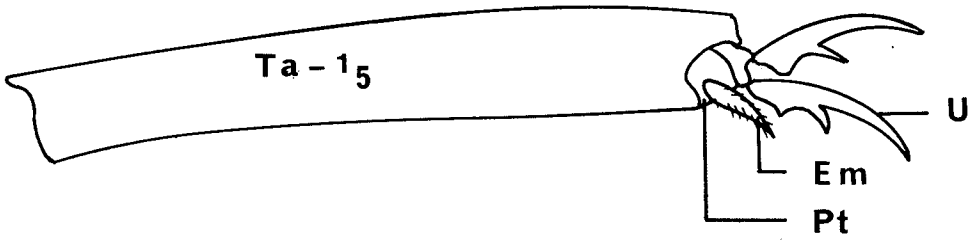
Dorsal view of the abdomen of the adult females of *A. mediolineata*, *A. campestris*, and *A. dorsalis*.



A. mediolineata Ludlow. A. campestris Dyar and Knab. A. dorsalis (Meigen).

Figure 9

Lateral view of the claw of an adult female (foreleg). Arrows indicate the points used to compare the size and shape of different claws. Em, empodium L_1, overall length of claw. L_2, length of the secondary tooth. L_3 length of the primary tooth. Pt, post-tarsus. PT, primary tooth. ST, secondary tooth. Ta-1_5, 5th tarsal segment of foreleg. U, claw.


Ta - 1 5
U
Em
Pt

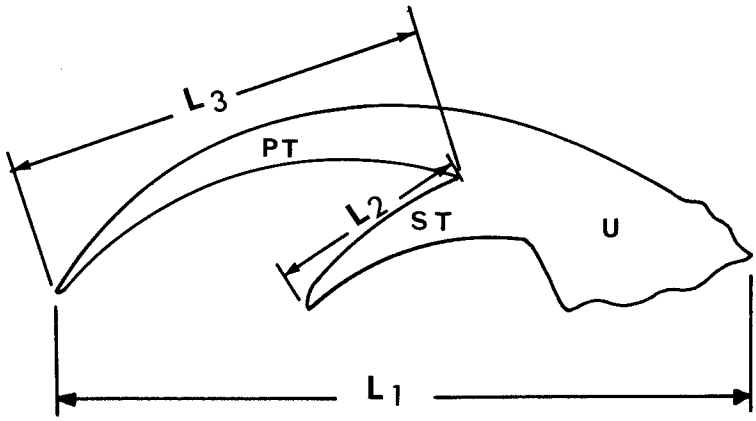

L3
PT
L2
ST
U
L1

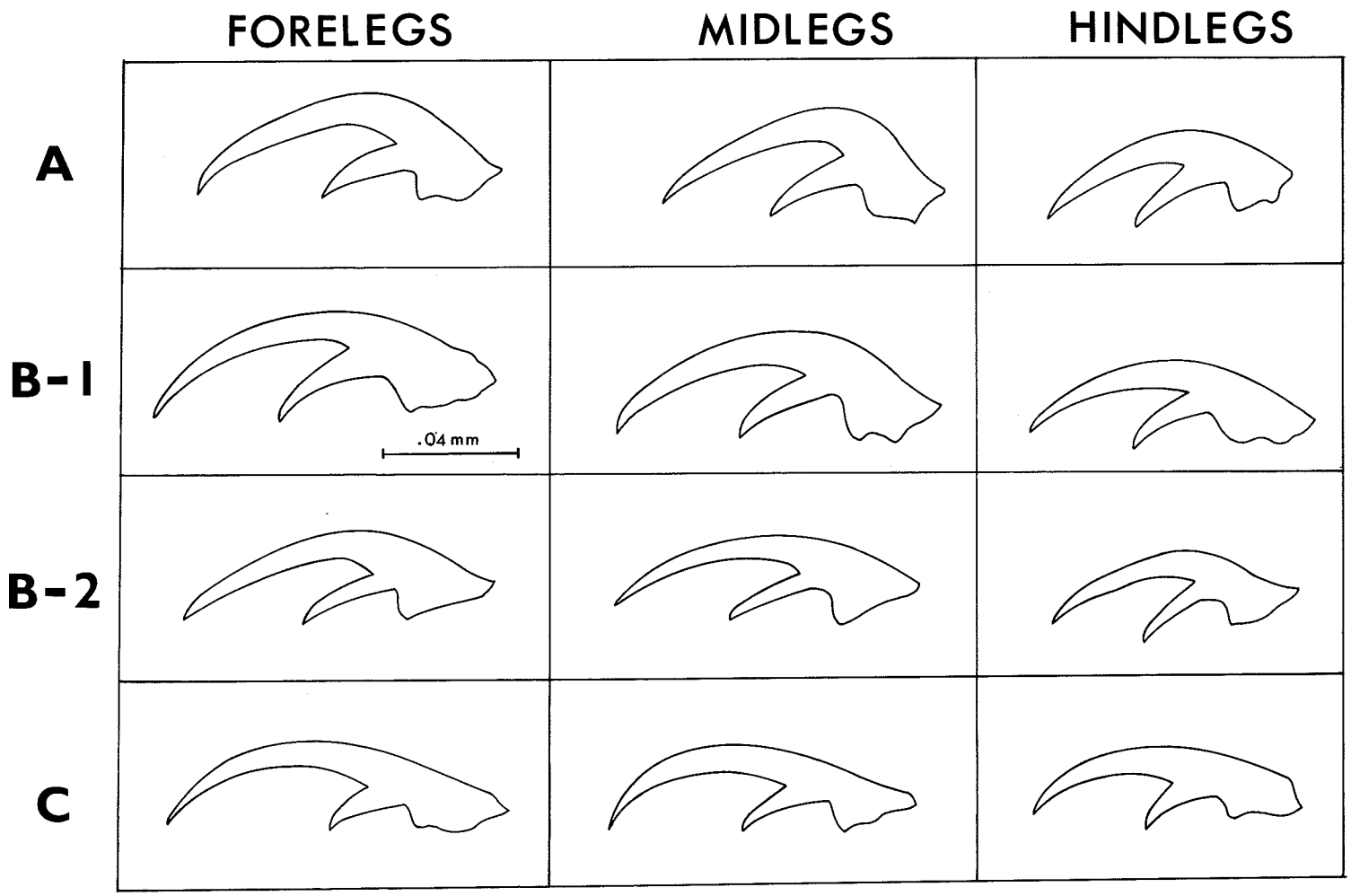
Figure 10

Claws of adult females of (A) *A. mediolineata* (S. Dos Palos, California).

(B-1) *A. campestris* (Churchill)

(B-2) *A. campestris* (Saskatoon)

(C) *A. dorsalis* (Saskatoon).


FORELEGS
MIDLEGS
HINDLEGS
A
B-1
B-2
C
.04 mm

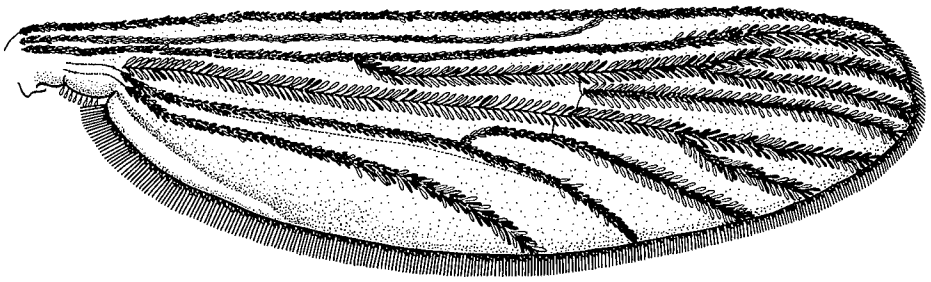
predominantly, with dark scales scattered on each vein. Distal end of anal vein almost entirely white. Usually no predominance of dark scales on vein R_{4+5} (Fig. 11). Length (from base to apex) 4.3-5.0 mm ($\bar{x}$ 4.8).

MALE IMAGO. Coloration very similar to that of female imago. Palpi as long as or slightly longer than proboscis. Antennae plumose; apical segment long and pilose, the rest short, inserted with hair-whorls; hairs long and dense, brownish-yellow. Wings narrower than those of female. Body length 5.8-7.3 mm ($\bar{x}$ 6.6). Claws of male imago are similar in shape to those of female except the anterior claws in pro- and mesothoracic legs. Metathoracic claw ratio 1.3:0.78 (Fig. 12B). Terminalia: The diagram of the male terminalia of an *Aedes* mosquito is shown in Fig. 13. In *A. campestris*, ninth tergal lobe as long as broad; each lobe bears 5-8 ($\bar{x}$ 5.9) stout spines. Phallosome conical, longer than broad, closed dorsally and open ventrally. Basistyle clothed with scales and numerous short and long setae; length about 3.4 times width at the middle (Fig. 14C). Apical lobe broadly rounded, inconspicuous, running uniformly down to the base with numerous dorsal setae. Basal lobe rounded, somewhat conical, protuberant, clothed with long, slightly curved, similar setae: a series of coarse spines (3-4) present at the basal margin (Fig. 14A). Dististyle slender, pilose, slightly swollen medially, with a few setae distally; about 0.6 times as long as basistyle. Dististylar claw slightly curved at distal end, 0.24 times as long as dististyle. Claspette stem curved, slender, pilose, bearing 3-5 ($\bar{x}$ 4) subapical setae; length 1.2 mm. Claspette filament about as long as claspette stem (length 1.2 mm), slender, slightly swollen medially, sickle-shaped (Fig. 14B).

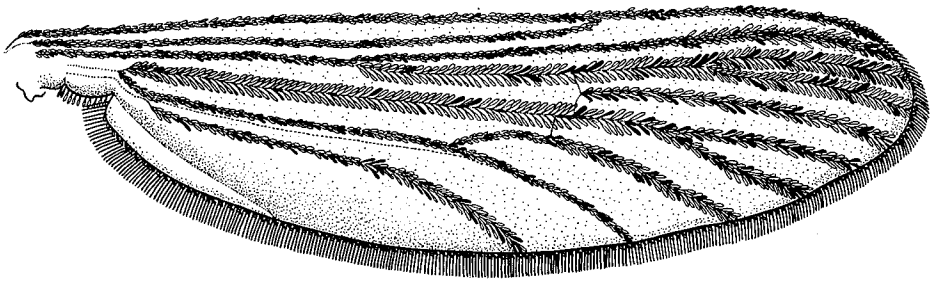
Figure 11

Pattern of the wing scales in adult females of *A. mediolineata*, *A. campestris* and *A. dorsalis*.

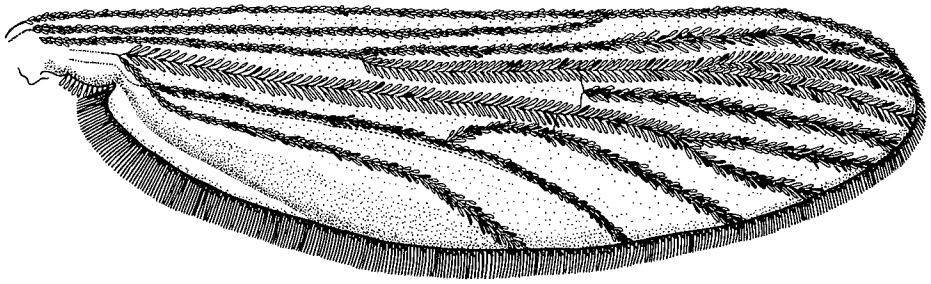
1mm



A. mediolineata Ludlow.



A. campestris Dyar and Knab.



A. dorsalis (Meigen).

Figure 12

Anterior and posterior claws (AU and PU, respectively) of adult males of

(A) *A. mediolineata*

(B) *A. campestris*

(C) *A. dorsalis*

Each pair of claws drawn to scale.

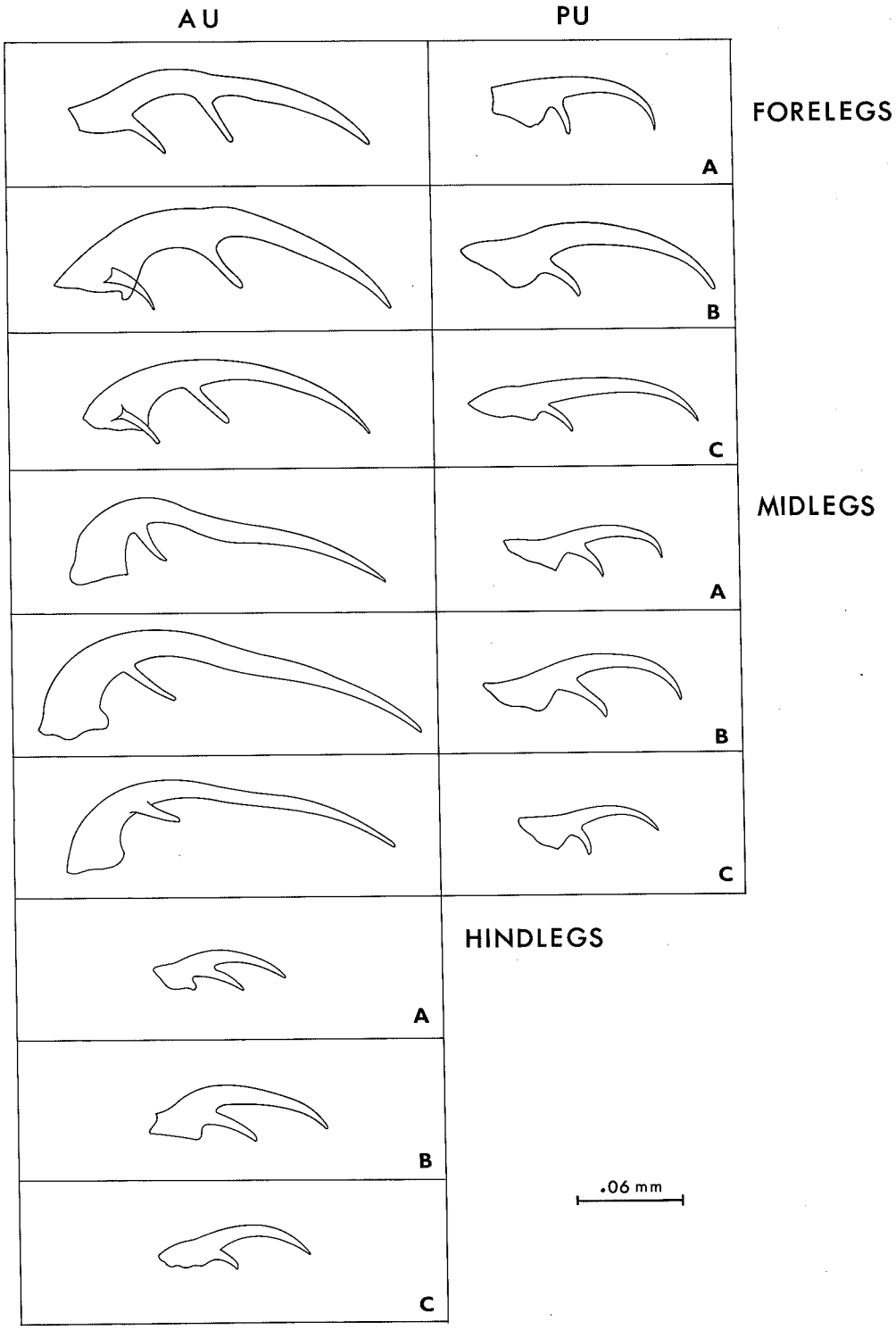

AU
PU
FORELEGS
A
B
C
MIDLEGS
A
B
C
HINDLEGS
A
B
C
.06 mm

Figure 13

Diagram of male terminalia of an *Aedes* mosquito, dorsal view.

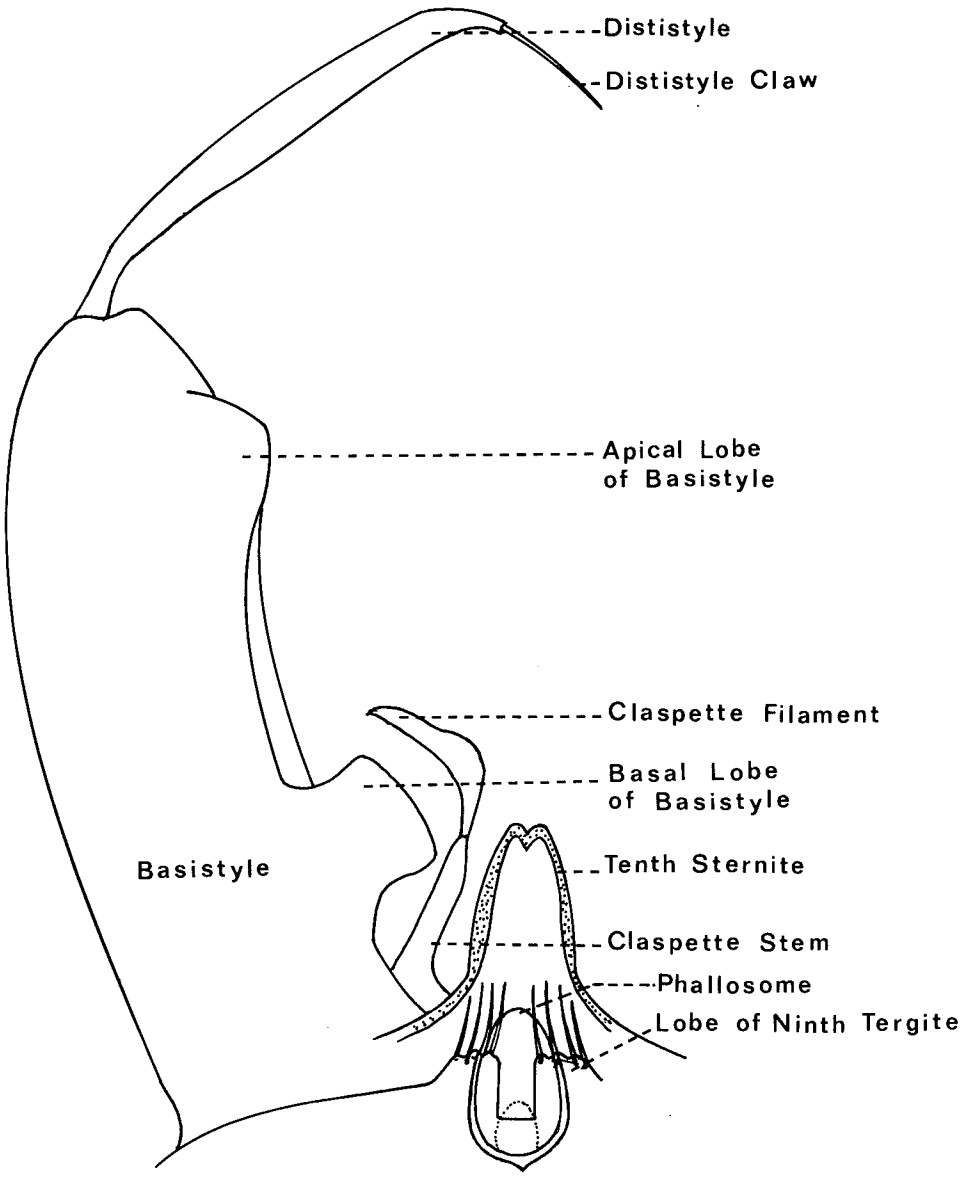

Dististyle
Dististyle Claw
Apical Lobe
of Basistyle
Claspette Filament
Basal Lobe
of Basistyle
Basistyle
Tenth Sternite
Claspette Stem
Phallosome
Lobe of Ninth Tergite

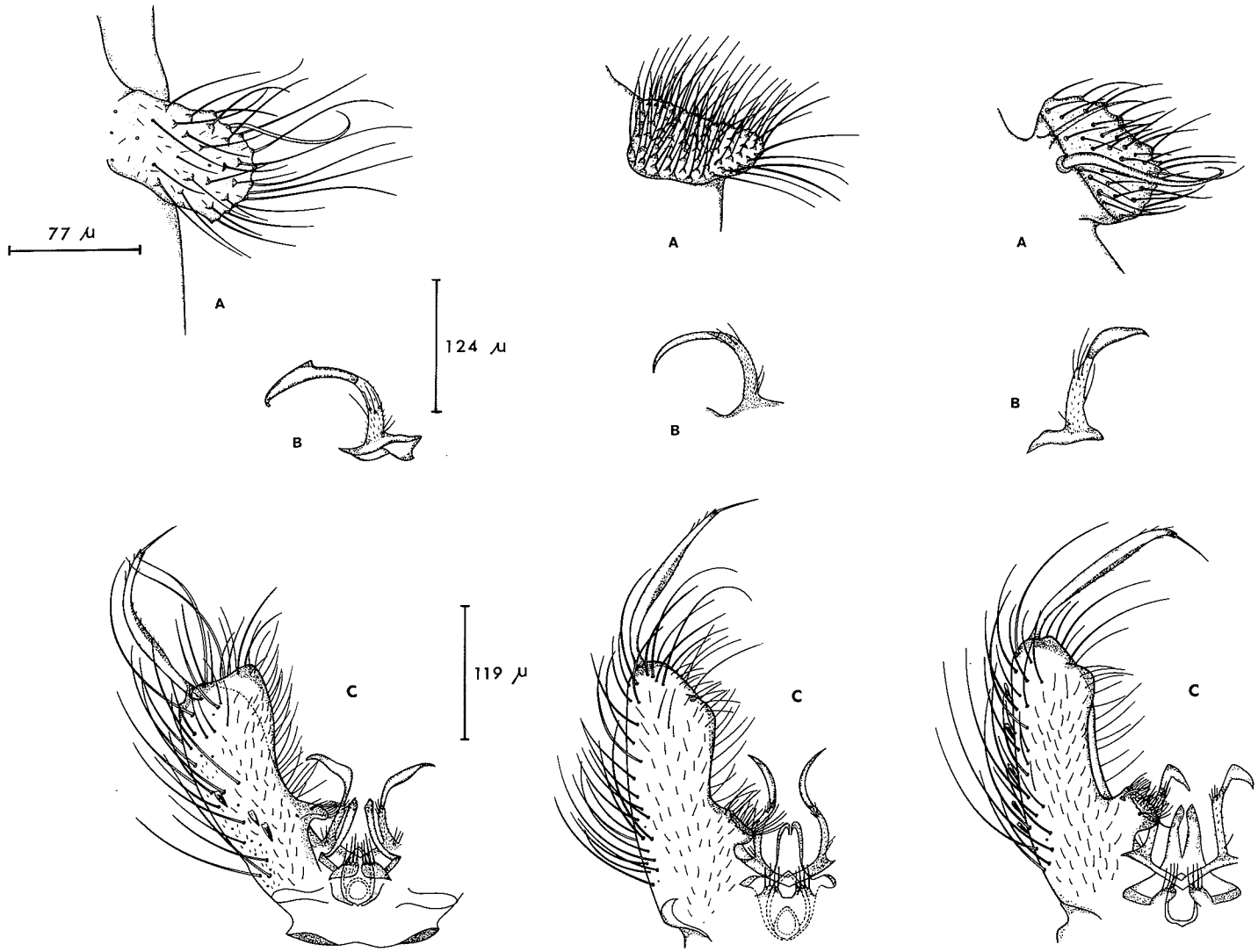
Figure 14

Male terminalia of *A. mediolineata*, *A. campestris*, and *A. dorsalis*.

(A) basal lobe of basistyle.

(B) claspette.

(C) male terminalia, dorsal view.



A. mediolineata Ludlow. A. campestris Dyar and Knab. A. dorsalis (Meigen).

A. Basal lobe

B. Claspette

C. Male terminalia

FOURTH-INSTAR LARVA. The fourth-instar larva of an *Aedes* mosquito is illustrated in Fig. 15. In *A. campestris*, Head: Rounded, wider than long (Fig. 16B); head capsule width 1.2-1.3 mm ($\bar{x}$ 1.2). Antenna slender, slightly tapering at apex, spinose and shorter than head; antennal tuft 6-11 branches ($\bar{x}$ 9), barbed, inserted only a little below the middle of antenna, not reaching to apex; at apex 2 digits and 3 hairs, two of which are inserted a little below the apex. Head setae: 4-C small, 2-5 branches ($\bar{x}$ 4); 5-C slightly barbed, 2-3 branches; 6-C usually single, slightly barbed; 7-C barbed, 8-15 branches ($\bar{x}$ 11), almost reaching to the middle of the antenna; 8-C single; 9-C double; 10-C single. Submentum 16-20 teeth ($\bar{x}$ 17.8). Prothoracic setae: 1-P long, slightly barbed, usually 2 branches, rarely 3 branches; 2-P short, single, nearly half as long as 1-P; 3-P short, double, about half as long as 1-P; 4-P short, usually single; 5-P long, barbed, triple; 6-P long, barbed, single; 7-P long, barbed, triple. Mesothoracic setae: 1-M long, 3 branches, about as long as head hair 5. Abdomen: Setae 6-I to 6-V usually 2 branches; seta 6-VI usually double (1-2), rarely single. Comb segment VIII with 15-33 ($\bar{x}$ 23.8) scales arranged in triangular patch (Fig. 17A); individual scales spatulated, broad and round, median spine slightly longer than lateral spines (Fig. 17B). Pentad seta 3-VIII about 7-11 branches ($\bar{x}$ 9). Siphonal index (length of the siphon, excluding the acus and valves, compared to its diamater at the base) 2.3-3.2 ($\bar{x}$ 3.0). Attached acus present. Pecten (Fig. 17C) 17-29 teeth ($\bar{x}$ 23.03) extended beyond basal half of siphon, 1-3 (usually 1) distal teeth detached. Siphonal seta 1-S, 5-8 branches ($\bar{x}$ 6), barbed, inserted beyond pecten about two-thirds from base of siphon, as long as or somewhat shorter than width of siphon at point of insertion. Siphonal seta 2-S single, stout, shorter than apical

Figure 15

Anatomical features of the 4th-instar larva of an *Aedes* mosquito: (A), (C) Dorsal and ventral aspects of head and thorax. (B) Submentum. (D) Dorsal and ventral aspects of abdominal segment I-VI. (E) Lateral aspect of abdominal segments VIII-X. A, antenna. 1-A, antennal tuft. C, head. 4-C, postclypeal hair. 5-C, upper head hair. 6-C, lower head hair. 7-C, preantennal hair. 8-C, inner occipital hair. 9-C, outer occipital hair. 10-C, dorsal eye hair. P, prothorax. 1 to 7-P, prothoracic setae. M, mesothorax. 1 to 8-M, mesothoracic setae. T, metathorax. Segment VIII: cs, comb-scales; and 1 to 5-VIII pentad setae. Segment X: ap, anal papillae; c.t., cratal hair tufts; pc.t, precratal hair tufts; sa, saddle; 1-X, lateral setae; 2-X, upper caudal setae; 3-X, lower caudal seta; 4-X, cratal and precratal setae. Siphon (S): ac, acus; p, pecten; 1-S, siphonal setal tuft, 2-S, dorsal preapical seta; 8-S, ventral apical seta.

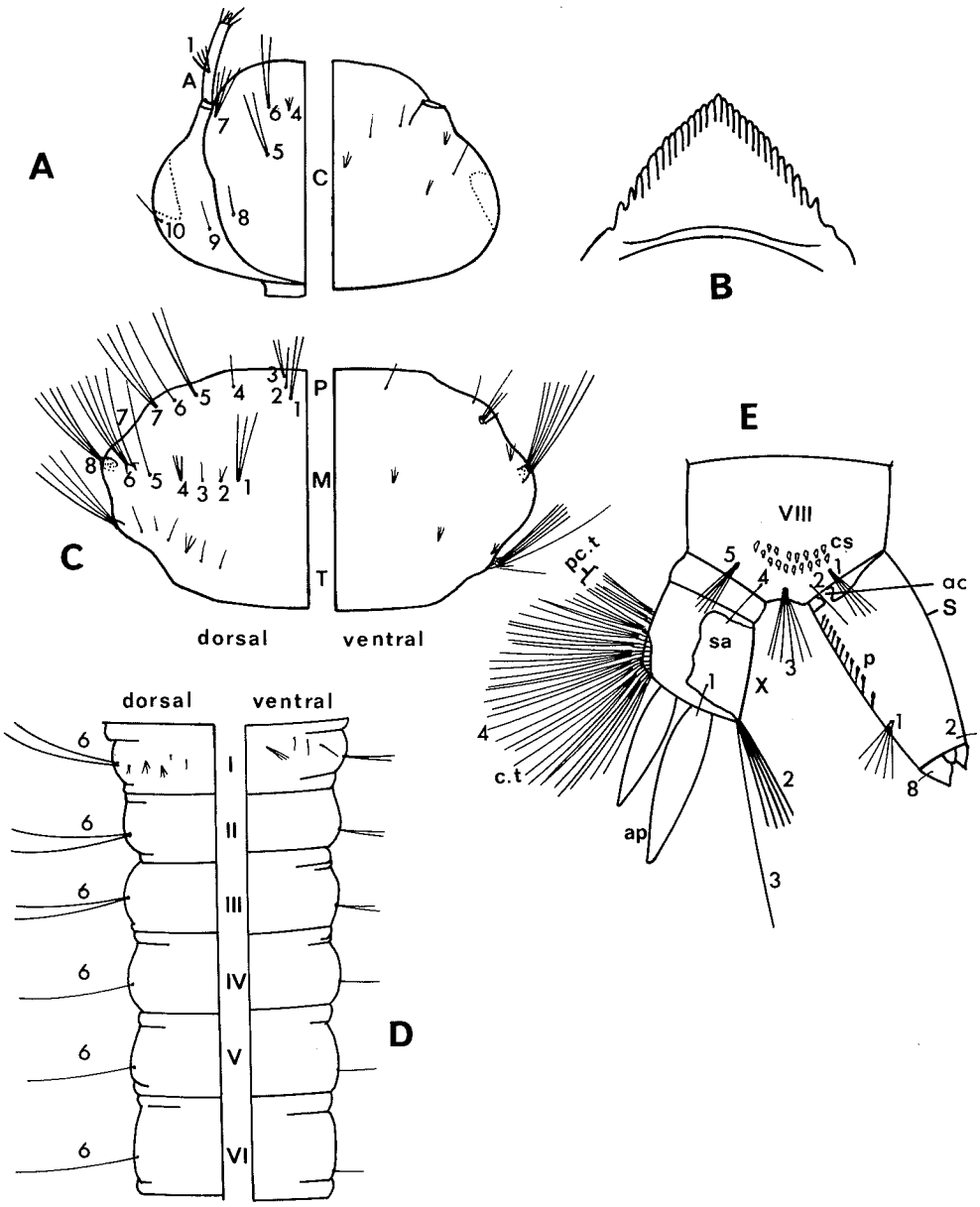
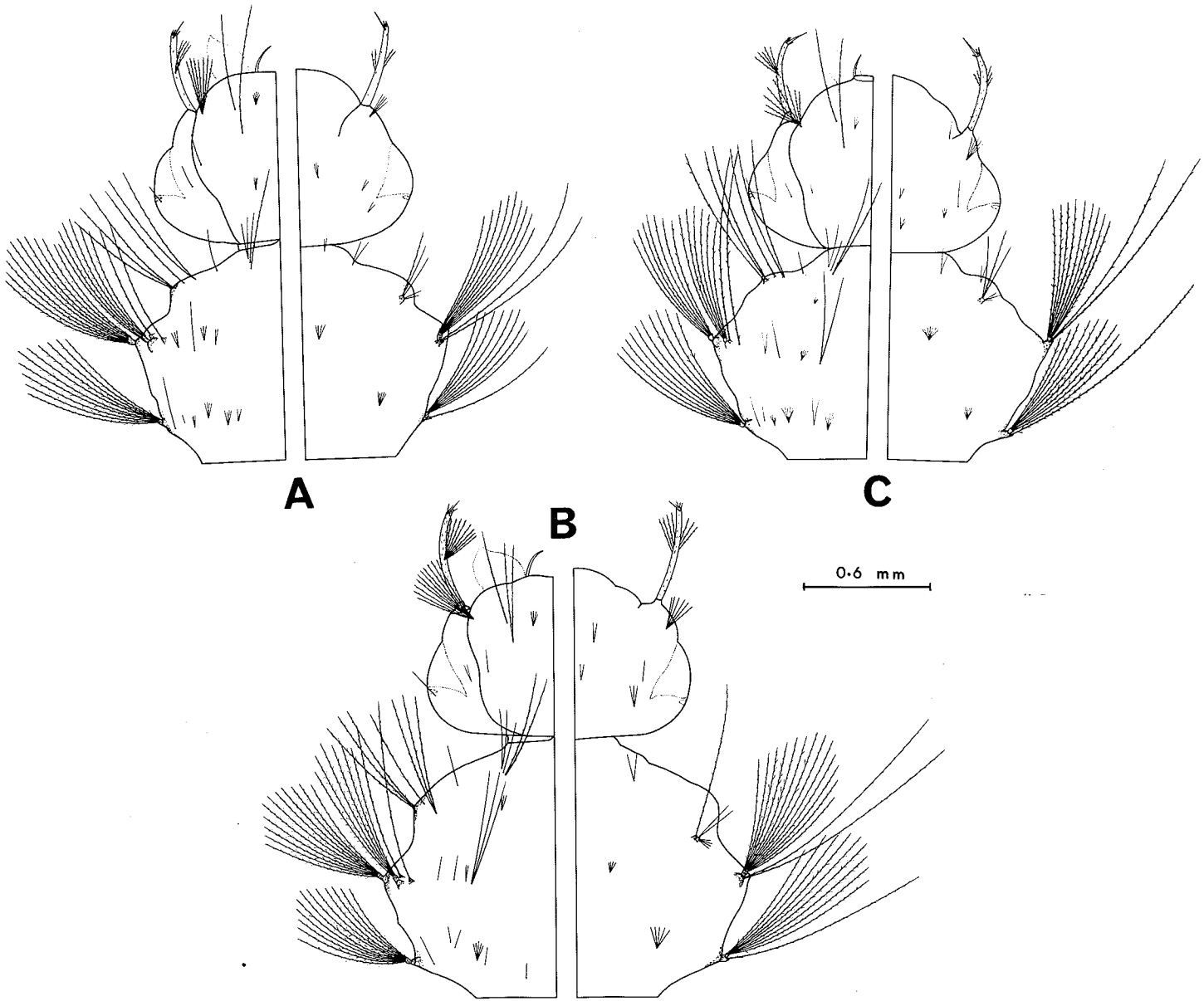

A
B
C
D
E
dorsal
ventral
P
M
T
C
A
VIII
cs
ac
S
sa
X
p
pc.t
c.t
ap
I
II
III
IV
V
VI

Figure 16

Dorsal view of the head and thorax of the 4th-instar larva of (A) *A. mediolineata*. (B) *A. campestris*. (C) *A. dorsalis*.


A
B
C
0·6 mm

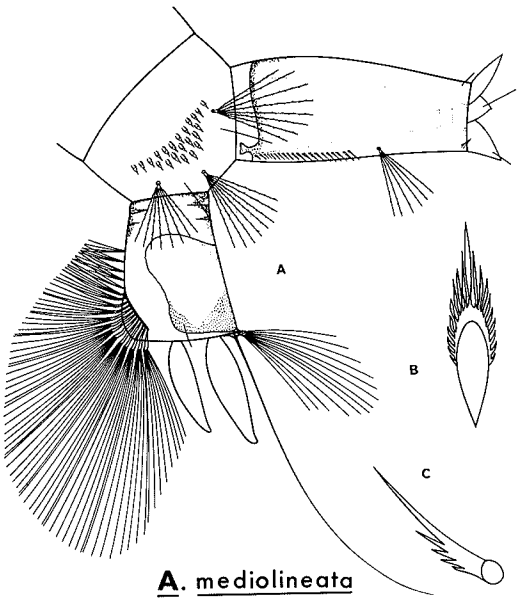
pecten tooth, inserted at least its length before apex of siphon. Anal segment longer than broad. Saddle extended 0.6-0.8 (two-thirds) down sides of segment X. Dorsoapical part of saddle spiculate. Seta 1-X single, shorter than saddle. Seta 2-X, 10-16 branches ($\bar{x}$ 13). Seta 3-X single, long, stout. Seta 4-X consisted of 15-20 cratals ($\bar{x}$ 16.5), and 3-5 ($\bar{x}$ 3.8) precratal tufts; each tuft bearing 7-12 branches ($\bar{x}$ 10). Anal papillae, short, bud-like, as long as or slightly shorter than the length of saddle (at pH 8.1-8.4); lower papillae shorter than upper papillae. The larvae reared in laboratory at pH 7 have elongate and gradually tapered anal papillae, each is about 3.6 times the saddle length.

FIRST-INSTAR LARVA. Head: hairs single; upper, lower and postclypeal hairs on each side being in straight line (Fig. 18A). Antennal ratio (ratio of length of antenna to the length of the mid-line of the head) approximately 1:2 (ca. 1:1.9). Antennal tuft triple, rarely double ($\bar{x}$ 2.96), inserted lower than the middle of antenna. Antennal hairs 2, 3, and 4, long and of equal length (Fig. 18B). Antenna lightly spiculate. Terminal segments (Fig. 18C): Comb scales (Fig. 18D) 5-8 ($\bar{x}$ 6.24), usually six or seven. Pentad hairs 2, 3, 4, of the same length; hair 3 slightly stouter than others; hair 1 and hair 5 about the same length, longer than one-half of hair 3. Siphonal ratio (length of the siphon to its width at the point of insertion of the siphonal hair), 3:1 (ca. 3.09:1). Pecten teeth (Fig. 18E) 3-5 ($\bar{x}$ 4.2). Siphonal hair single, inserted beyond pecten teeth and reaching beyond apex of siphon.

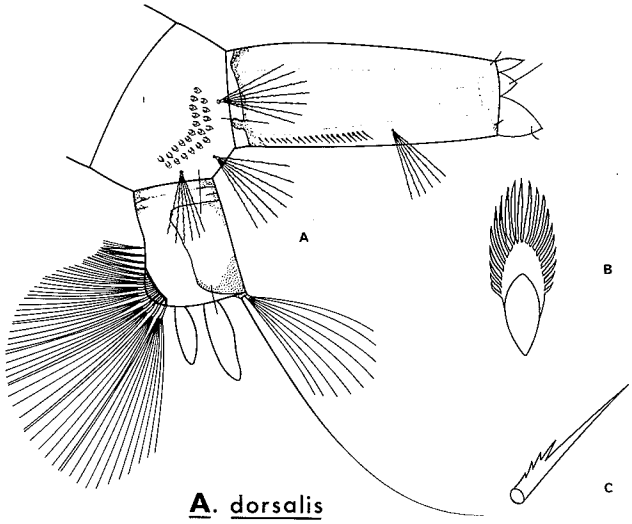
DISTRIBUTION. North America, in northern prairie regions (Barr, 1958) and western semiarid plains of Canada and U.S.A. (Carpenter and LaCasse, 1955).

Figure 17

Lateral aspect of abdominal segments VIII- X of the 4th-instar larvae of *A. mediolineata*, *A. campestris*, and *A. dorsalis*. (A) Terminal abdominal segments. (B) comb scales. (C) pecten.



A. mediolineata



A. dorsalis

A. campestris

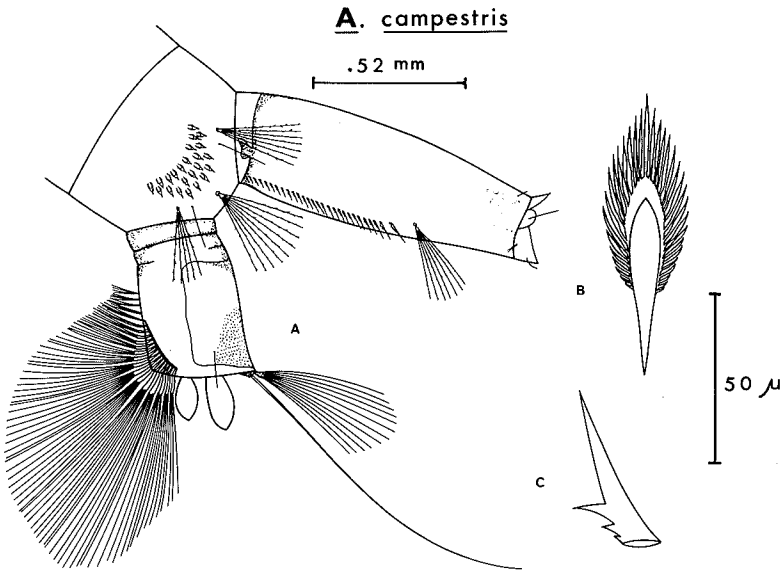
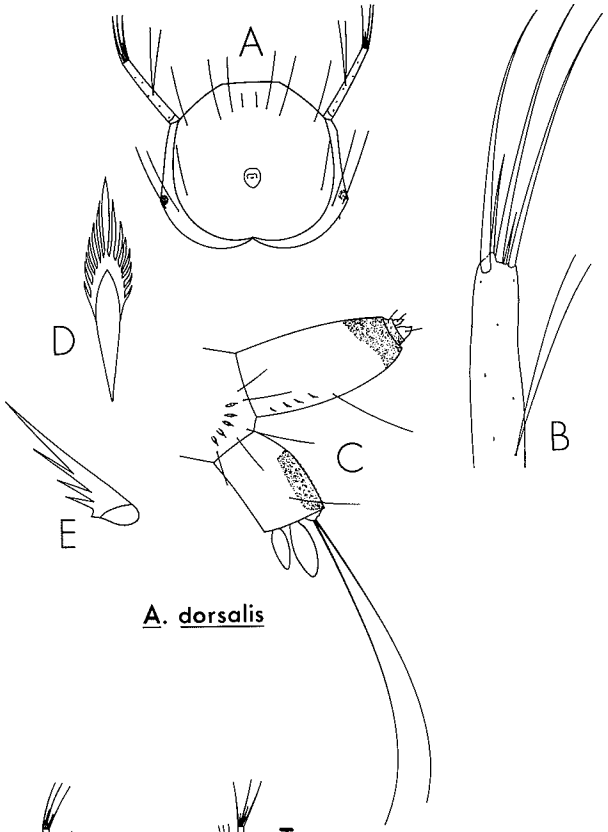
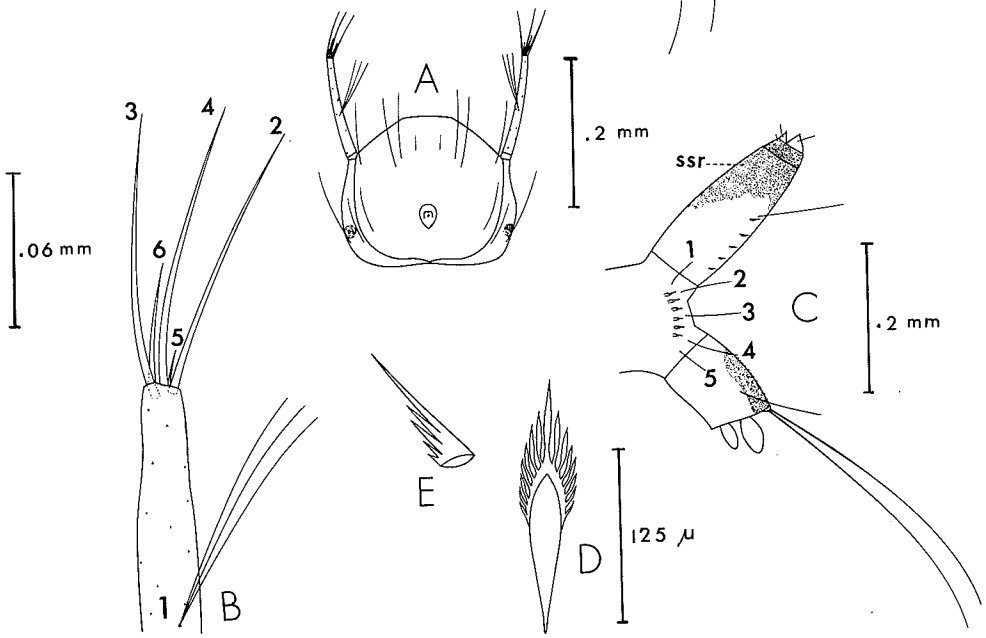


Figure 18

First-instar larvae of *A. campestris* and *A. dorsalis*. (A) Head. (B) Terminal antennal appendages: 1, antennal hair tuft; 2, inner subapical hair; 3, outer subapical hair; 4, dorsal median terminal hair; 5, hyaline process; 6, dorsal external finger process. (C) Terminal segments: 1-5, pentad hairs; ssr, secondary sclerotized ring. (D) Comb scales. (E) Pecten tooth.



A. dorsalis



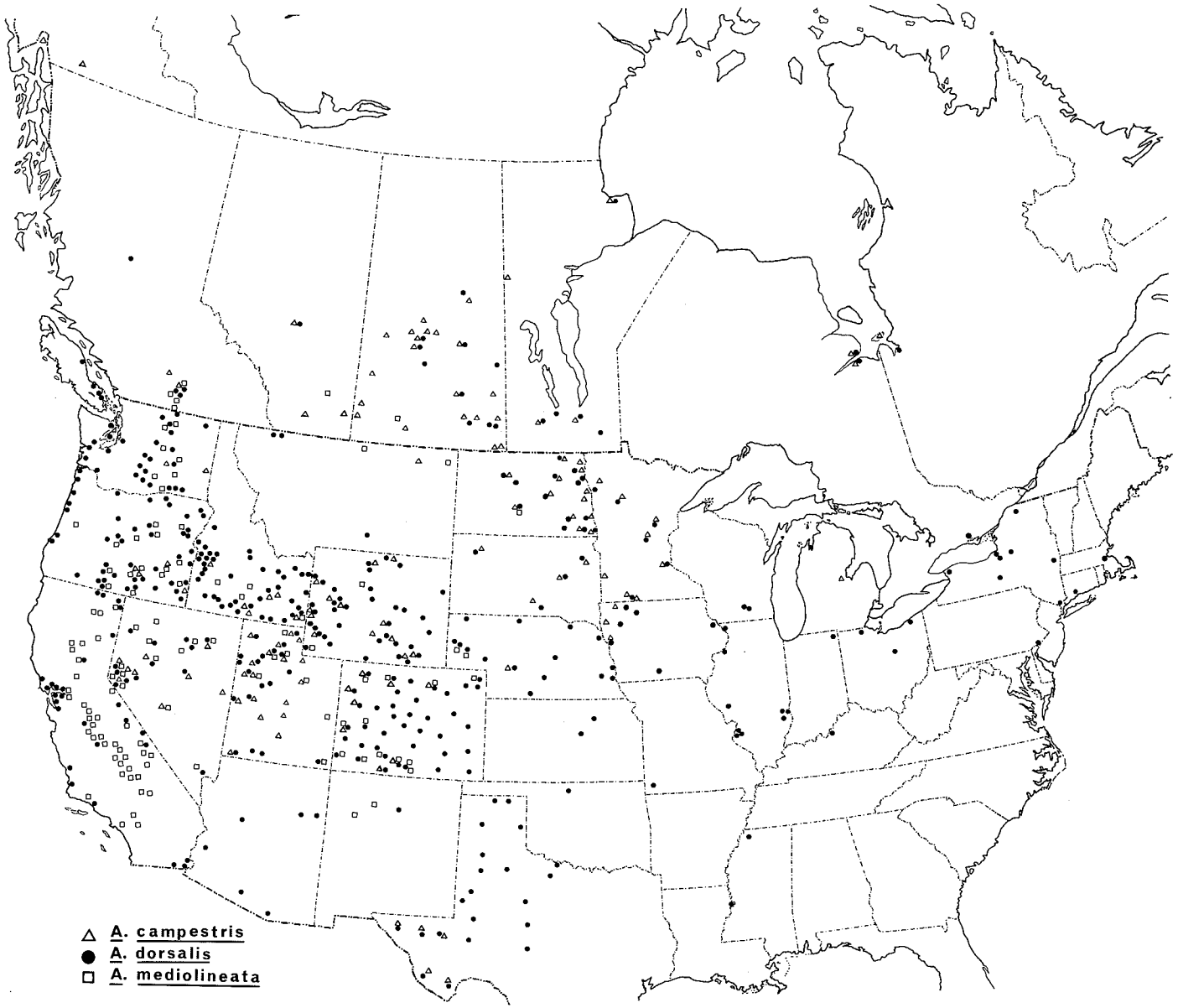
A. campestris

CANADA. (Fig. 19). Alberta (Twinn, 1949) - Edmonton, Lethbridge, Medicine Hat (Rempel, 1950). Letham (Ellis, R., U. of M. Coll.) British Columbia (Twinn, 1949) - Clinton (Meredith and Phillips, 1973). Kamloops (Kiceniuk and Phillips, 1974). Manitoba (Twinn, 1949) - Churchill (Twinn *et al.*, 1948; Vockeroth, 1954). Flin Flon (Kalpage and Brust, 1968). Hudson's Bay (Barr, 1958). Winnipeg (McLintock, 1944). Ontario (Carpenter and LaCasse, 1955) - Charlton Island, Moose Factory, and Moosonee (Steward and McWade, 1961). Quebec (Twinn, 1949). Saskatchewan (Twinn, 1949) - Aberdeen, Aneroid, Arran, Borden, Cedoux, Dafoe, Dundurn, Fox Valley, Kenosee, Kindersley, Lang, Maple Creek, Rosthern, Saskatoon, Scott, Stockholm, White Fox (Rempel, 1953). Oxbow (Stone *et al.*, 1959). Quill Lakes, Regina (Rempel, 1950). Estevan (Present study). Yukon Territory (Barr, 1958) - Whitehorse (Vockeroth, 1954; Stone *et al.*, 1959).

UNITED STATES (Fig. 19). Alaska (Matheson, 1929) - Skagway (Dyar, 1922). Colorado (Carpenter and LaCasse, 1955) - Alamosa Co.: Alamosa, Del Norte, Conejos Co.: La Jara; Garfield Co.: Rifle, Mesa Co.: Grand Junction; Moffat Co.: Elk Springs, Maylell; Rio Blanco Co.: Rangely; Routt Co.: Heyden; Weld Co.: Nunn, Roggen (Harmston and Lawson, 1967). Idaho (Gjullin and Eddy, 1972) - Ada, Bannock, Bear Lake, Butte, Cassia, Franklin, Oneida, Power (Gjullin and Eddy, 1972). Iowa (Carpenter and LaCasse, 1955) - Plymouth Co.: Akron; Osceola Co.: Sibley; Monona Co.: Whiting (Rowe, 1942). Michigan (Barr, 1958) - Midland Co. (Irwin, 1941). Minnesota (Barr, 1958) - Clearwater, Crow Wing Co. (Owen, 1937). East Grand Forks (Dyar, 1922), Jackson Co., Lincoln Co., Morrison Co., Polk Co., and Ramsey Co. (Owen, 1937). The Twin Cities (Barr, 1958). Montana (Mail, 1934) - Glasgow (Dyar, 1922). Nebraska (Tate and Gates, 1944) - Lincoln Co. (Tate and Gates, 1944).

Figure 19

Geographical distribution of *A. campestris*, *A. dorsalis*, and *A. mediolineata* in North America (North of Mexico).


△ A. campestris
● A. dorsalis
□ A. mediolineata

Nevada (Chapman, 1966) - Churchill Co.: Fallon; Big Soda Lake (Richards *et al.*, 1956; Chapman, 1966). Douglas Co.: Genoa (Chapman, 1966). Elko Co.: Halleck (Chapman, 1966). Eureka Co.: Eureka (Chapman, 1966). Lyon Co.: Lahonton Reservoir, Wabuska (Chapman, 1966). Mineral Co.: Schurz (Richards *et al.*, 1956). Nye Co. (Richards *et al.*, 1956). Washoe Co.: Gerlach, Reno (Chapman, 1966). White Pine Co.: Baker (Richards *et al.*, 1956). North Dakota (Dyar, 1922) - Buxton, Cavalier, Devils Lake, Fargo, Grafton, Hankinson, Mandan, Milnor, Minot, New Rockford, Niles Siding, Northwood, Ramsey Co., St. Thomas, and Valley City (Post and Munro, 1949). Oregon (Carpenter and LaCasse,1955) - Malheur, Harney, and Lake (Gjullin and Eddy, 1972). South Dakota (Gerhardt, 1966) - Beadle Co., Brule Co., Codington Co., and Perkins Co. (Gerhardt, 1966). Texas (McGregor and Eads, 1943) - Brewster, Culberson, El Paso, Hudspeth, Jeff Davis, Loving, Pecos, Presidio, Reeves, Terrell, Ward, and Winkler (McGregor and Eads, 1943). Utah (Dyar, 1922) Box Elder Co.; Cache Co.: Logan; Davis Co. (Richards *et al.*, 1956). Garfield Co. (Dyar, 1922). Juab Co.: Fish Springs, Trout Creek; Millard Co.: Clear Lake, Garrison; Salt Lake Co. (Richards *et al.*, 1956). Sevier Co.: Elsinore (Dyar, 1922). Tooele Co., and Weber Co. (Richards *et al.*, 1956). Duchesne Co., Rich Co., Sanpete Co., Sevier Co., Summit Co. (Nielsen and Rees, 1961). Washington (Carpenter and LaCasse, 1955) - Grant, Whitman (Gjullin and Eddy, 1972). Wisconsin (Carpenter and LaCasse, 1955). Wyoming (Owen and Gerhardt, 1957) - Albany Co.: Albany, Laramie; Big Horn Co., Greybull; Carbon Co.: Arlington, Rawlins, Seminoe Dam; Lincoln Co.: Afton, Auburn, Diamondville; Sheridan Co.: Sheridan; Sublette Co.: Boulder, Daniel (Owen and Gerhardt, 1957).

Aedes (Ochlerotatus) dorsalis (Meigen)

Culex dorsalis Meigen, 1830, Syst. Beschr. Zweifl. Ins., 6:242.

Culex curriei Coquillett, 1901, Can. Ent., 33:259.

Culex onondagensis Felt, 1904, N.Y. Sate Mus. Bull., 79:278.

Culex lativittatus Coquillett, 1906, Ent. News, 17:109.

Aedes quaylei Dyar and Knab, 1906, Jour. N. Y. Ent. Soc., 14:191.

Grabhamia broquetti Theobald, 1913. Entomologist, 46:154.

Aedes grahami Ludlow, 1920, Ins. Ins. Mens., 7:154.

FEMALE IMAGO. Medium-sized, length 5.0-6.2 mm ($\bar{x}$ 5.51).

Head: Proboscis black, with pale scales scattered on the basal part. Palpi short, one-sixth as long as proboscis, speckled with pale scales. Antennae with subequal joints, rugose, pilose, dark-brown. Torus yellowish-brown, bears short, broad, white scales on dorsal and inner surfaces. Clypeus dark-brown, bell-shaped. Vertex with short, narrow, curved, white scales; and a series of white setae. Occiput dark, covered with curved, narrow, whitish scales and erect, forked, whitish scales in a broad median zone; a small submedian patch with curved, narrow, dark-brown scales and a few dark, erect, forked scales; posterior margins of eyes lined with a series of dark bristles; lateral zone with short, broad, white scales and some dark speckle.

Thorax: Anterior pronotal lobes elliptical, covered with dark-brown bristles at the tip; with narrow, curved, whitish scales below and scattered with white setae. Scutum integument dark; broad median stripe with short, narrow, curved, yellowish-brown or dark-brown scales and posterior end with forked-like process, stripe varies in width; lateral margins dark brown; remainder of thorax with narrow, curved, whitish scales, (Fig. 7C). Posterior pronotum with narrow, dark-brown scales on dorsal half and ventrally covered with white scales. Scutellum

trilobed, yellow or yellowish-brown, covered with narrow, curved, whitish scales and silver setae along the lobes. Postnotum cup-shaped, prominent, nude, yellowish-brown to dark-brown. Pleura with scattered broad, white scales. Mesepisternal scale patch with white scales extending to anterior angle and continuing to prealar area. Mesepimeral scale patch with scales extending to lower margin. Hypostigma covered with scales. Lower mesepimeral bristles 1-5 ($\bar{x}$ 3.4). Abdomen: Dorsal appearance with median patch of sordid-white scales and two patches of dark scales on each segment (Fig. 8). First tergite with median patch of broad white scales and many long, white hairs. Venter with white scales. Remaining tergites white scales surrounded by solid, dark, irregular, submedian patches with produced T-shape marking on segments 2-5; dark patches on segments 6, 7 sprinkled with white scales. Cerci black. Sternite white scales with dark scales speckled on middle area. Legs: Femur: anterior surface with white and dark scales intermixed; posterior surface creamy white. Tibia white, speckled with dark scales. Tarsi: first tarsal segment white, speckled with dark scales. Foretarsi: dark with narrow, white basal and apical bands on segments 1 and 2; segment 3 with narrow, white basal band; segments 4 and 5 dark. Midtarsi: segment 1, 2, 3 with narrow, white basal and apical bands; segment 4 dark with small basal band; segment 5 dark. Hindtarsi: segments 1-3 with narrow basal and apical white bands; basal band on segment 4; segment 5 white with very few black scales scattered. Tarsal claws: primary tooth long, slightly curved; secondary tooth short, divergent. Primary tooth is about 3.3 times as long as secondary tooth (Fig. 10C). Claw ratio 1.36:0.99. Wings (Fig. 11): white scales predominate, with dark scales intermixed. Dark scales often predominate on vein R_{4+5}; length 3.96-5.0 mm

($\bar{x}$ 4.56).

MALE IMAGO. Coloration similar to that of female imago. Palpi as long as or slightly longer than proboscis. Antennae plumose; apical segment long and pilose; the others short, with long and dense brown hairs. Body length 5.8-7.1 mm ($\bar{x}$ 6.5). Claws: shape of claws is similar to that in female except for anterior claws of pro- and mesothoracic legs (Fig. 12C). Metathoracic claw ratio 1.3:0.7. Terminalia (Fig. 14C): Ninth tergal lobe as long as broad; each lobe bears 3-7 short, stout spines ($\bar{x}$ 5.6). Phallosome conical, longer than broad, closed dorsally and open ventrally. Basistyle slender, clothed with scales and numerous short and long setae; length 4.2 times width at middle. Apical lobe of basistyle rounded, inconspicuous, bearing numerous dorsal setae. Basal lobe rounded, constricted at the base and expanded apically which forms prominent knob; bearing coarse setae on median surface; basal margin bears a large, strong recurved spine and a short stout spine (Fig. 14A). Dististyle slender, pilose, slightly broadened medially, 0.6 times as long as basistyle. Dististylar claw about 0.22 times as long as dististyle. Claspette stem almost straight stem, pilose, bearing 4-5 subapical setae. Claspette filament shorter than claspette stem, swollen medially on convex side, distal portion gradually tapers to a blunt, recurved point (Fig. 14B).

FOURTH-INSTAR LARVA. Head rounded, wider than long (Fig. 16C); head capsule width 1.16-1.22 mm ($\bar{x}$ 1.19). Antenna spinose, slender, about half as long as head; antennal tuft 3-7 branches ($\bar{x}$ 5.04), barbed, inserted near the middle of antenna, not reaching the apex. Head setae: 4-C small, 2-4 branches ($\bar{x}$ 2.6); 5-C long slightly barbed, single, occasionally double; 6-C single, barbed; 7-C barbed, 5-12

branches ($\bar{x}$ 6.9), almost reaching the insertion of antennal tuft; 8-C single; 9-C single; 10-C single. Submentum 18-23 teeth ($\bar{x}$ 19.3). Prothoracic setae: 1-P long, slightly barbed, usually double; 2-P short, single, less than half as long as 1-P; 3-P short, slightly longer than 2-P, double, rarely 1 or 3 branches; 4-P short, single; 5-P long, barbed, 2 branches, rarely 3; 6-P long, single, barbed; 7-P long, barbed, 3 branches. Mesothoracic setae: 1-M long, 2 branches, slightly smaller than head hairs. Abdomen: Setae 6-I to 6-IV usually double or triple; setae 6-V and 6-VI double. Comb on segment VIII 12-31 scales ($\bar{x}$ 23.02) arranged in triangular patch (Fig. 17A); individual scales spatulated, broad and round, median spine equal to lateral spines (Fig. 17B). Pentad seta 3-VIII about 6-16 branches ($\bar{x}$ 9.5). Siphonal index 2.2-3.1 ($\bar{x}$ 2.6). Acus present. Pecten (Fig. 17C) 15-24 teeth ($\bar{x}$ 18.67), confined at the basal half of siphon, with evenly spaced teeth. Siphon seta 1-S, 4-9 branches ($\bar{x}$ 6), barbed, inserted beyond the middle of siphon but less than two-thirds of the tube; setae much shorter than the width of siphon at point of insertion. Siphon seta 2-S single, shorter than apical pecten tooth. Anal segment longer than broad. Saddle extended about 0.52-0.65 ($\bar{x}$ 0.63) or one-half to three-fifths down the sides of segment X. Dorsoapical part of saddle spiculate. Seta 1-X single, much shorter than saddle. Seta 2-X multiple, 10-19 branches ($\bar{x}$ 15). Seta 3-X single, long and stout. Seta 4-X consists of 11-17 cratal ($\bar{x}$ 14.9), and 2-4 precratal tufts ($\bar{x}$ 2.6); each tuft 7-10 branches ($\bar{x}$ 8.6). Anal papillae short, bud like, 1.13 times the length of saddle at water pH 8.1-8.4.

FIRST-INSTAR LARVA. Head: Hairs single; upper, lower, and postclypeal hair on each side in a straight line (Fig. 18A). Antennae very slightly spiculate, about half as long as head. Antennal ratio 1:2. Antennal hairs 2, 3, 4 of the same length (Fig. 18B). Antennal tuft 2-3 branches ($\bar{x}$ 2.6) inserted near the middle of antenna. Terminal segment (Fig. 18C): Comb scales (Fig. 18D) ranging from 4-7 ($\bar{x}$ 5.5), usually 5 or 6. Pentad hairs 2, 3, 4 of about the same length; hair 3 slightly longer than the others. Siphonal ratio 2:1 (ca. 1.93:1). Pecten (Fig. 18E) 3-5 ($\bar{x}$ 3.58). Siphonal hair single; inserted beyond pecten, and reaching the apex of siphon.

DISTRIBUTION. Australia, Central Asia, China, Denmark, England, Europe, Finland, Germany, Hungary, North China, North Russia, Mongolia, Siberia (Marshall, 1938). United States and Canada (Carpenter and LaCasse, 1955).

CANADA (Fig. 19). Alberta (Carpenter and LaCasse, 1955) - Edmonton (Rempel, 1950). British Columbia (Twinn, 1949) - Duncan (Hearle, 1926). Fraser Delta (Curtis, 1967). White Rock to Caulfields (Rempel, 1950). Kamloops, Douglas Lake, Merritt, Qualicum, Saratoga Beach, Cowichan Bay (Can. Nat. Coll.). Manitoba (Twinn, 1949) - Brandon (S. Pierce Coll. 1937, Univ. of Man.). Churchill, Portage la Prairie (Present Study), Winnipeg (McLintock, 1944; Kalpage and Brust, 1968). Ontario (Twinn, 1949) - Belleville (James *et al.*, 1969). Moose Factory, Moosonee (Steward and McWade, 1961). Windsor area (Benedict, 1962). Quebec (Twinn, 1949) - Rupert House (Vockeroth, 1954). Saskatchewan (Twinn, 1949)- Carlyle district (Rempel, 1950). Dafoe, Dundurn, Kisbey, Nipawin, Regina, Yellow Grass, and Yorkton (Rempel, 1953). Estevan and Saskatoon (Present Study).

UNITED STATES (Fig. 19). Arizona (Richards *et al.*, 1956) - Apache Co.: Springerville, Lyman Dam, St. Johns; Navajo Co.: Joseph City; Pima Co.: Tucson; Yavapai Co.: Cornville; Yuma Co.: Parker, Yuma (Richards *et al.*, 1956). Santa Cruze (McDonald *et al.*, 1973). California (Freeborn and Bohart, 1951) - Goose Lake, Guadalupe Lake, Honey Lake, Laguna Lake, and Tule Lake (Bohart, 1956). Antelope, Niles, Thrall (Dyar, 1922). Solano Co.: Suisan, Vallejo, and Dixon (Bohart, 1954). Yolo Co. (Telford, 1958). Colorado (Dyar, 1922) - Florissant (Dyar, 1922). Specimens were collected in 40 of the 53 counties (Harmston and Lawson, 1967). Connecticut (Matheson, 1945) - New Haven (Matheson, 1945). Delaware (Carpenter and LaCasse, 1955). Idaho (Dyar, 1922) - Ada, Adams, Bannock, Bear Lake, Bingham, Blaine, Bonneville, Butte, Camas, Canyon, Caribou, Cassia, Custer, Elmore, Franklin, Fremont, Gem, Gooding, Jefferson, Jerome, Lincoln, Madison, Minidoka, Oneida, Owyhee, Payette, Power, Teton, Twin Falls, Washington (Gjullin and Eddy, 1972). Boise (Stone *et al.*, 1959). Market Lake (Dyar, 1922). Illinois (Dyar, 1922) - Chicago (Dyar, 1922; Ross, 1947). Dupo (Adams and Gordon, 1943). Cahokia, Camp Grant, Chanute Field, Dupo, East Moline, East St. Louis, Great Lake Naval Training Station, Oak Park, Riverdale, Savanna, Scott Field, Summit (Ross, 1947). Indiana (Silverly, 1972) - Delaware, Steuban (Silverly, 1972). Iowa (Rowe, 1942) - Buena Vista Co.: Marathon, Storm Lake;Clarke Co.: Osceola; Dickinson Co.: Lake Park; Dubuque Co.: Dubuque; Monona Co.: Onawa, Whiting; Palo Alto Co.: Ruthven (Rowe, 1942). Kansas (Carpenter and LaCasse, 1955) - Camp Phillips, Fort Riley (Olson and Keegan, 1944). Kentucky (Quinby *et al.*, 1944) - Jefferson (Quinby *et al.*, 1944). Louisiana (Dyar, 1922) —Delta (Dyar, 1922; King *et al.*, 1960).

Massachusettes (Dyar, 1922) - Boston (Dyar, 1922). Minnesota (Dyar, 1922) - Clearwater, Itasca State Park (Price, 1960). Crow Wing, Jackson, Morrison, Polk, Ramsey (Owen, 1937). East Grand Forks (Dyar, 1922). Mississippi (Miles and Rings, 1945) - POW Camp, Como (Miles and Rings, 1945). Missouri (Smith and Enns, 1968) - Camp Crowder, Jefferson Barracks (Olson and Keegan, 1944). Montana (Dyar, 1922) - Glacier Park, and Milk River (Mail, 1934). Laurel (Dyar, 1922). Nebraska (Dyar, 1922) - Lincoln (Dyar, 1922). Bridgeport, Lexington, Lincoln, McCook, Nebraska City, Niobrara, North Platte, Omaha, Ord, Oshkosh, Scottsbluff, Valentine, Walthill, Whitney (Tate and Gates, 1944). Scotts Bluff Co.: Mitchell (Edmunds,1958); Blair, Dakota City, Peru, Plattsmouth, Rulo, Tekamah (Rapp, 1958). Nevada (Dyar, 1922) - Churchill Co.: Fallon, Hazen, Soda Lake; Clark Co.: Bunkerville, Glendale, Mesquite, Moapa, Overton; Douglas Co.: Genoa, Minden; Elko Co.: Carlin, Deeth, Elko, Lamoille, Midas, Owyhee, Wilkins; Eureka Co.: Beowawe, Dunphy, Eureka, Palisade; Humboldt Co.: Golconda, Winnemucca; Lander Co.: Austin, Battle Mt.; Lincoln Co.: Alamo, Caliente, Panaca; Lyon Co.: Dayton, Fernley, Hazen, Lahontan Reservoir, Wabuska; Mineral Co.: Schurz: Nye Co.: Gabbs (Chapman, 1966). New Mexico (Dyar, 1922) - North East New Mexico (Ferguson and McNeel, 1954); Pecos (Dyar, 1922). New York (Dyar, 1922) - Albany Co.: Albany; Niagara Co.: Fort Niagara; Oneida Co.: Utica; Onondaga Co.: Baldwinsville, Syracuse; Queens Co.: Flushing; Rockland Co.: Orangeburg; Tomphins Co.: Ithaca (Barnes *et al.*, 1950). St. Lawrence Co.: Canton (Doll, 1970). North Dakota (Dyar, 1922) - Butte, Casselton, Cass Co., Cavalier, Fargo, Grand Forks, Hankinson, Jamestown, LaMoure Co., Litchville, Mandan, Milnor, Minot, Minto, New Rockford, Northwood, Oberon, Towner, Traill Co., Valley City, Walsh Co.; Ward Co. (Post

and Munro, 1949). Devils Lake (Dyar, 1922). Ohio (Carpenter and LaCasse, 1955) - Lake Co., Lucas Co., and Wayne Co. (Venard and Mead 1953). Oklahoma (Carpenter and LaCasse, 1955) - Great Salt Plains near Cherokee; Wide spread near Gypsum Hills region of Oklahoma (Rozeboom, 1942). Oregon (Dyar, 1922) - Baker, Clatsop, Coos, Crook, Deschutes, Grant, Hoo River, Harney, Jackson, Jefferson, Klamath, Lake, Lincoln, Malheur, Morrow, Tillamook, Umatilla, Union, Wallowa, Wheeler (Gjullin and Eddy, 1972). Klamath Falls (Dyar, 1922). Pennsylvania (Dyar, 1922) - Philadelphia (Dyar, 1922). South Dakota (Carpenter and LaCasse, 1955) - this species is present throughout S. Dakota (Gerhardt, 1966). Texas (Rueger and Druce, 1950) - Baylor Co., Brewster Co., Culberson Co., Dickens Co., Gaines Co., Hale Co., Hansford Co., Hudspeth Co., Kimble Co., Lubbock Co., Midland Co., Ochiltree Co., Potter Co., Presidio Co., Reeves Co., Runnels Co., Shackelford Co., Terry Co., Upton Co., and Wheeler Co. (Eads *et al.*, 1951). Wichita Co.: Sheppard Field (Rueger and Druce, 1950). Utah (Dyar, 1922) - Grand Junction (Dyar, 1922). Juab Co.: Fish Springs, Gandy, Juab, Yuba Dam; Kane Co.: Glendale; Millard Co.: Burbank, Clear Lake, Fairbanks, Garrison, Lyndyl; San Juan Co.: Bluff; Toocle Co.: Dolomite, Dugway, Grantswille, Salt Springs, Wendover; Washington Co.: St. George; common and widespread in the following counties: Box Elder, Cache, Davis, Salt Lake, Utah, Weber (Richards *et al.*, 1956). Washington (Dyar, 1922) - Oroville, West Seattle (Dyar, 1922). Adams, Benton, Chelan, Cowlitz, Douglas, Grant, Grays Harbor, Jefferson, King, Kittitas, Kitsap, Mason, Okanogan, Pacific, Stevens, Thurston, Walla Walla. Yakima (Gjullin and Eddy, 1972). Wisconsin

(Dyar, 1922) - Madison (Dyar, 1922). Dane Co.; Hope Lake near Cambridge (Dickinson, 1944). Wyoming (Carpenter and LaCasse , 1955) - Albany Co.: Albany, Bosler, Laramie, Woods Landing; Big Horn Co.: Basin, Greybull; Carbon Co.: Arlington, Baggs, Elk Mountain, Saratoga, Siminoe Dam; Converse Co.: Glenrock, Orin; Fremont Co.: Dubois, Shoshoni; Hot Springs Co.: Thermopolis; Lincoln Co.: Afton, Auburn, Border, Cokeville, Diamondville, Kemmerer, Opal; Natrona Co.: Casper; Platte Co.: Wheatland; Sheridan Co.: Ranchester, Ucross; Sublette Co.: Big Piney, Boulder, Daniel, Pinedae; Sweetwater Co.: Farson, Green River; Teton Co.: Jackson, Wilson; Uinta Co.: Evanston, Urie; Weston Co.: Newcastle (Owen and Gerhardt, 1957).

Aedes (Ochlerotatus) mediolineata (Ludlow)

Grabhamia mediolineata Ludlow, 1907, Can. Ent., 39:129.

Aedes melanimon Dyar, 1924, Ins. Ins. Mens., 12:126

Aedes klotsi Matheson, 1933, Proc. Ent. Soc. Wash., 35:69.

FEMALE IMAGO. Medium sized species, length 4.4-5.7 mm ($\bar{x}$ 5.19). Head: Proboscis dark-brown with speckle of pale scales at the basal half up to two-thirds of the length of proboscis. Palpi short, brownish-black, with pale scales at the tip and central area. Palpi about one-fifth as long as proboscis. Antennae brown, slender, with subequal joints, rugose, pilose. Torus light to dark-brown, with white scales on dorsal and inner surfaces. Clypeus conical, nude, brown or dark-brown. Vertex with short, narrow, curved, whitish scales; and a series of long, silver setae. Occiput dark-brown, with

narrow, curved, white scales and erect, forked whitish scales on broad median zone; submedian zone with narrow, curved, brown scales and dark, erect, forked scales intermixed; hind edge of eyes with a series of brown bristles; lateral zone with broad, white scales. Thorax: Anterior pronotal lobe elliptical, pilose dorsally, covered with golden bristles at the tip; the lower part covered with broad, white scales and sprinkles of golden setae. Scutum integument dark-brown; broad median stripe and lateral margin with short, curved, narrow, brown or dark-brown scales (usually of copper shade), the middle area of stripe spotted with narrow, curved, white scales; remainder of thorax covered with narrow, white scales at the anterior part, and brown patched posteriorly (Fig. 7A). Posterior pronotum with narrow, dark-brown scales on dorsal half and ventrally covered with white scales. Scutellum trilobed, yellowish-brown integument, with narrow, curved, white scales and golden setae along the lobe. Postnotum cup-shaped, nude, prominent, yellowish- or reddish-brown. Pleura heavily clothed with very broad white scales. Mesepisternal scale patch extending to anterior angle and continues to prealar area. Mesepimeron heavily covered with broad, white scales extending to lower margin. Lower mesepimeral bristles 1-7 ($\bar{x}$ 3.3). Hypostigma with scales. Abdomen: First tergite with median patch of broad white scales and a series of long silver hairs. Dorsal appearance of segments 2-7, solid brown scales, with white basal band on each segment, bands extend wider at lateral sides; central area with dorso-median white stripe (Fig. 8). Cerci black. Sternites white scaled, with irregular dark patches on median and submedian areas. Legs: Femur: white and dark scales intermixed on anterior surface, posterior surface white

scaled. Tibia with white scales speckled with dark scales on anterior surface. Tarsi: first tarsal segment white and dark scales intermixed. Foretarsi dark, with basal and apical white bands on segments 1 and 2; segment 3 with basal band; segments 4, 5 entirely dark. Midtarsi: segments 1, 2, and 3 with basal and apical white bands; segment 4 with basal band; segment 5 entirely dark. Hindtarsi: segments 1-3, dark with basal and apical white bands; segment 4 dark with basal band; segment 5 white. Tarsal claws: claws are abruptly curved with produced prominent angle on the dorsum of primary tooth (Fig. 10A). Secondary tooth long and almost parallel to primary tooth. Primary tooth is about 2.1 times the length of secondary tooth. Claw ratio 1.28: 0.79. Wings (Fig. 11): dark-brown and white scales intermixed, with dark scales predominating. Costa dark-brown scaled at the anterior margin, a series of white scales at the base of posterior margin of the vein. Subcosta with brown scales but lighter shade scales predominate. First, second, and third longitudinal veins, dark scales predominate, with few light scales. Fourth longitudinal vein brown and white scales intermixed, dark scales predominate at the distal end. Veins 5 and 6 almost entirely dark. Length 4.1-5.2 mm ($\bar{x}$ 4.48).

MALE IMAGO. Coloration similar to that of female imago. Palpi are slightly longer than proboscis. Antennae plumose; apical segment long, pilose; the rest short, inserted with long, dense, brownish-yellow hair-whorls. Wings narrower than those of female. Body length 5-6.8 mm ($\bar{x}$ 6.04). Metathoracic claw ratio 1.1:0.7. Terminalia (Fig. 14C): Ninth tergal lobe flattened about as long as broad; each lobe bears 4-8 stout spines ($\bar{x}$ 5.5). Phallosome conical, round at

apex, open ventrally. Basistyle clothed with scales and numerous short and long setae; length 4.25 times width at middle. Apical lobe large, prominent, with short setae on dorsal surface. Basal lobe quadrate, prominent, with apex projecting upward; central area clothed with a few fine setae; margin of basal lobe with one short spine and one long spine recurved distally.(Fig. 14A). Dististyle slender, about 0.6 times (two-thirds) as long as basistyle. Dististylar claw 0.22 times as long as dististyle. Claspette stem curved, slender, pilose, bearing 3-4 setae subapically. Claspette filament with long shank and sharply expanded near the middle, blade-like, longer than stem, distal portion curved (Fig. 14B).

FOURTH-INSTAR LARVA. Head rounded, wider than long (Fig. 16A); head capsule width 1-1.16 mm ($\bar{x}$ 1.07 mm). Antenna slender, spinose, shorter than head; antennal tuft 5-9 branches ($\bar{x}$ 6.9), barbed, inserted a little below the middle, not reaching to apex; at apex 2 digits, and 3 hairs with 2 smaller hairs inserted a little below apex. Head setae: 4-C small, multiple 2-4 branches ($\bar{x}$ 3); 5-C slightly barbed, single; 6-C single, slightly barbed; 7-C barbed, 6-10 branches ($\bar{x}$ 7.7), reaching to the middle of antenna; 8-C single; 9-C single; 10-C single. Submentum 21-25 teeth ($\bar{x}$ 22.93). Prothoracic setae: 1-P long, slightly barbed, usually single, occasionally double; 2-P short, single, less than half as long as 1-P; 3-P small, double, as long as 2-P or slightly shorter; 4-P short, single; 5-P long, barbed, usually double, occasionally triple; 6-P long, single; 7-P long, triple, rarely 4 or 5. Mesothoracic setae: 1-M very small, double. Abdomen: Setae 6-I to 6-V usually double, occasionally triple; 6-VI usually single. Comb segment VIII 19-27 scales ($\bar{x}$ 22.46), arranged in triangular patch

(Fig. 17A); individual scales spatulated, with prominent long median spine (Fig. 17B). Pentad seta 3-VIII multiple 6-11 branches ($\bar{x}$ 9.09). Siphonal index ranged from 2.3-2.9 ($\bar{x}$ 2.61). Acus present. Pecten (Fig. 17C) ranged from 16-25 teeth ($\bar{x}$ 20.8), confined at basal half of siphon. Siphon seta 2-S single, stout, short, about half as long as apical pecten tooth, inserted near apex of siphon. Anal segment longer than broad. Saddle extended 0.7 (three-quarters) down side of segment X. Dorsoapical part of saddle spiculate. Seta 1-X single, shorter than saddle. Seta 2-X about 10-20 branches ($\bar{x}$ 12.6). Seta 3-X long, single. Seta 4-X consisted of 11-17 cratal ($\bar{x}$ 14.7), and 4-5 precratal tufts ($\bar{x}$ 4.3), each tuft 7-13 branches ($\bar{x}$ 10.3). Anal papillae elongate and gradually tapered, about as long as or slightly longer than the length of saddle; lower papillae shorter.

FIRST-INSTAR LARVA. The first-instar larva of *A. mediolineata* has been described by Dodge (1966) under the name *A. melanimon*. The larvae are very similar to *A. dorsalis* with 5-7 comb scales; 5-6 pecten teeth; and gills half as long as anal segment.

DISTRIBUTION*. Western United States, North America and Eurasia (Bohart, 1956).

CANADA (Fig. 19). Alberta (Burgess, 1957) - Brooks (Burgess, 1957). British Columbia (Present Study) - Kamloops, Nicola, Merritt, Osoyoos (Can. Nat. Coll.). Saskatchewan (Holmberg and Trofimenkoff, 1968). Swift Current (Holmberg and Trofimenkoff, 1968; McLintock's

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* Prior to this study *A. mediolineata* was known as *A. melanimon*.

Coll., 1967).

UNITED STATES (Fig. 19). California (Barr, 1955) - Kern, Modoc, Los Angeles (Richards, 1956). Feather, Kern, Pit, San Joaquin, Santa Ana, Santa Inez (Bohart, 1956). Porterville, Tulare (Barr, 1955). Solano Co., Yolo Co. (Telford, 1958). Specimens from the U. S. Nat. Museum, Bohart's coll. (1916-1950), Miura's Coll. (1970), were collected from Antelope Valley, Bakersfield, Emery, Kings Co., Princeton, Co., S. dos Palos, Stanislaus Co., Tulare Co., and Woodland Co. Colorado (Richards, 1956) - Alamosa, Costilla, Delta, Mesa, Moffat, Rio Grande, Routt, Weld (Richards, 1956). Costilla Co.: Ft. Garland; La Plata Co.: Durango; Misa Co.: Grand Junction; Montesuma Co.: Cortez; Sedgewick Co.: Jumbo Reservoir; Weld Co.: Greeley, Grover, Kuner, Plattville (Harmston and Lawson, 1967). Idaho (Gjullin and Eddy, 1972) - Blaine (Gjullin and Eddy, 1972). Specimens of U. S. Nat. Museum were collected from Berger, Bruneau, Hazelton, Montpelier, Roberts, and Shoshone. Montana Richards (1956) - Blaine (Richards, 1956). Chinook, Poplar (U.S. Nat. Museum). Nebraska (Richards, 1956) - Morill, Scottsbluff (Richards, 1956). Nevada (Richards, 1956) - Clark Co.: Bunkerville; Churchill Co.: Fallon; Douglas Co.: Minden City; Elko Co.: Lamoille Canyon, Carlin, Ruby Valley, Midas, Elko, Deeth, Owyhee; Eureka Co.: Beowawe, Dunphy; Humboldt Co.: Golconda Paradise Valley, Winnemucca; Lander Co.: Mattle Mt.; Lyon Co.: Dayton, Fernley, Smith, Yerington; Mineral Co.: Schurz; Ormsby Co.: Brunswick Canyon, Carson City; Persling Co.: Lovelock; Storey Co. (Truckee River); Washoe Co.: Reno, Sparks, Wadsworth, Gerlack (Chapman, 1966). New Mexico (Richards, 1956) - Sandoval, Valencia (Richards, 1956). North Dakota (Stone *et al.*, 1959) - Fort Lincoln (Stone *et al.*,

1959). Oregon (Gjullin and Eddy, 1972) - Baker Co., Crook Co., Deschutes Co., Grant Co., Klamath Co., Lake Co., Lane Co., Malheur Co., Morrow Co., Umatilla Co., Wheeler Co. (Gjullin and Eddy, 1972). Specimens of U. S. Nat. Museum were collected from McDermitt, Ft. Flamath, Silver Lake. Utah (Richards, 1956) - Cache Co.: Mendon; Davis Co., Juab Co.: Topaz; Salt Lake Co.: Salt Lake City; San Juan Co.: Bluff (Richards, 1956; Richards *et al.*, 1956). Duchesne Co., Grand Co., and Utah Co. (Nielsen and Rees, 1961). Washington (Richards, 1956) - Benton (Richards, 1956; Gjullin and Eddy, 1972). Franklin (Richards, 1956). Douglas, Grant, Okanogan, Yakima (Gjullin and Eddy, 1972). Wyoming (Richards, 1956) - Hot Springs (Richards, 1956).

Comparison of larval and adult characters

The counts and measurements of characters in fourth instar larvae, which are shown in Table 9 plus the characters mentioned in the description of species, were based on: 49 larvae of *A. campestris* from Churchill, Manitoba; 35 larvae of *A. campestris* from Saskatoon, Saskatchewan; 50 larvae of *A. dorsalis* from Saskatoon; and 39 larvae of *A. mediolineata* from Dos Palos, California. The first instar larval characters were based on 50 larvae of *A. campestris* from Churchill and Saskatoon; 50 larvae of *A. dorsalis* from Saskatoon. The adult characters of both sexes were based on at least 15 specimens of each species.

Table 9. Comparison of larval and adult characters in *A. campestris*, *A. dorsalis* and *A. mediolineata*.

CHARACTERS	SPECIES		
	A. campestris	*A. dorsalis*	*A. mediolineata*
		First-instar larva	
Upper head hair	1	1	-
Lower head hair	1	1	-
Antennal hair 1	2-3 ($\bar{x}$ 2.96±0.04)	2-3 ($\bar{x}$ 2.6±0.1)	-
Antennal ratio (Length of antenna/ length of midline of the head)	1:2	1:2	-
No. of comb scales	5-8 ($\bar{x}$ 6.24 ± 0.1)	4-7 ($\bar{x}$ 5.5±0.1)	5-7
Appearance of comb scales	median spine prominent	median spine prominent	-
No. of pecten teeth	3-5 ($\bar{x}$ 4.2±0.08)	3-5 ($\bar{x}$ 3.58±0.09)	5-6
Siphonal ratio	3:1	2:1	Siphon longer than in *A. dorsalis*
Sclerotized ring on siphon	Secondary sclerotized ring extends below middle	Secondary sclerotized ring confined to the distal half	-

Table 9. Cont.

Siphonal hair	Single, reaches beyond apex of siphon	Single, reaches only to apex of siphon	single, does not reach to apex of siphon
		Fourth-instar larva	
Antennal tuft	6-11 ($\bar{x}$ 9)	3-7 ($\bar{x}$ 5)	5-9 ($\bar{x}$ 6.9)
Upper head hair 5	2-3	1	1
Lower head hair 6	1	1	1
Submentum teeth	16-20 ($\bar{x}$ 17.8)	18-23 ($\bar{x}$ 19.3)	21-25 ($\bar{x}$ 22.9)
Prothoracic hair formula (branches on hairs 1 to 7)	212-1-313	212-1-213	112-1-213, occasionally 112-1-313.
Mesothoracic hair 1	3 branches, stouter and longer than in *A. dorsalis*	2 branches, long	2 branches very small
Lateral abdominal hairs 1-VI	2-2-2-2-2-2	2-2-2-2-2-2 occasionally 2-3 branches on segment 1-V	2-2-2-2-2-1
No. of comb scales	15-33 ($\bar{x}$ 24)	12-31 ($\bar{x}$ 23)	19-27 ($\bar{x}$ 22)
Appearance of comb scale	Median spine and lateral spines almost of equal length	Median spine and lateral spines of equal length	Median spine much longer than lateral spines

Table 9. cont.

No. of pecten teeth	17-29 ($\bar{x}$ 23), pecten extend beyond middle of siphon, usually 1 (0-3) distal tooth detached	15-24 ($\bar{x}$ 19), confined to basal half of siphon, teeth evenly spaced.	16-25 ($\bar{x}$ 21), confined to basal half of siphon, teeth evenly spaced.
Siphonal index	3.0	2.58	2.61
Siphonal hairs (tuft)	5-8 ($\bar{x}$ 6)	4-8 ($\bar{x}$ 6)	5-8 ($\bar{x}$ 6)
Saddle	Extends two-thirds of the way down each side of segment X	Extends one-half to 3/5 of the way down each side of segment X	Extends three-quarters of the way down each side of segment X.
No. of cratal tufts	15-20 ($\bar{x}$ 16)	11-17 ($\bar{x}$ 15)	11-17 ($\bar{x}$ 15)
No. of precratal tufts	3-5 ($\bar{x}$ 4)	2-4 ($\bar{x}$ 3)	4-5 ($\bar{x}$ 4)
		Female Imago	
Dorsum of thorax	Broad median stripe and lateral margins of thorax yellowish brown, remainder of thorax white, patches of brown scales on posterior end	Median stripe and narrow lateral margins on thorax yellowish brown, width of median stripe extremely variable, remainder of thorax white	Broad median stripe and lateral margins of thorax yellowish or reddish brown, central area of median stripe spotted with white scales, remainder of thorax white with brown patches on posterior end

Table 9. cont.

Pleural scales	Broad, dull white scales	Broad, white scales similar to those in *A. campestris*	White and very broad scales compared to those in *A. campestris* and *A. dorsalis*
Lower mesepimeral bristles	3-7 ($\bar{x}$ 4)	1-5 ($\bar{x}$ 3)	1-7 ($\bar{x}$ 3)
Abdominal tergites	Dark patches with scattered white scales on each segment.	White scales sprinkled on dark patches of segments 6 and 7 only	No white scales on dark patches
7th abdominal tergite	Almost entirely white except for a few dark scales	Mostly white scales with small patches of dark scales	Predominantly dark scaled
Wing scales	White and dark scales intermixed, predominantly white scales on all veins	White and dark scales intermixed, extremely variable, in some females veins R_{4+5} are predominantly dark scaled and in other females wing scales are like *A. campestris*	Predominantly dark scaled, distal end of each vein mostly dark scaled.
Costal vein	Predominantly white scaled	Predominantly white scaled	Predominantly dark scaled

Table 9. cont.

Tarsal claw ratio (length of primary tooth/length of basal portion of claw)	1.49:0.98	1.36:0.99	1.28:0.79
Ratio of primary tooth/ secondary tooth of claw	2.1:1	3.3:1	2.1:1
		Male Imago	
Metathoracic claw ratio	1.3:0.78 secondary tooth long, almost parallel to gradually curved primary tooth	1.3:0.7 secondary tooth short, divergent	1.1:0.7 secondary tooth long, almost parallel to abruptly curved primary tooth.
Apical lobe of basistyle	Round, inconspicuous	Round, not prominent	Large and prominent
Basal lobe of basistyle	Rounded, slightly raised numerous fine setae on median surface, setae longer than in *A. dorsalis*, large spines on basal margin absent	Rounded, constricted at base and raised, expanded apically basal margin with 1 large strong recurved spine and 1 short stout spine, setae on median surface shorter and coarser than in *A. campestris*	Quadrate, with apex projecting upward, margin of lobe with 1 short and 1 long narrow spine recurved distally, setae on median surface short with long setae along the margin

Table 9. cont.

Claspette filament	Sickle-like, gradually curved, as long as stem	Swollen medially on convex side, shorter than stem	Sharply expanded over the middle, longer than stem
Claspette stem	curved	almost straight	curved

KEY TO SPECIES

Fourth-instar larva

1. Upper and lower head hairs single, pecten with evenly spaced teeth and confined to the basal half of siphon, siphon length less than 3 times width at base (about 2.6 times). . . . (2)

   Upper head hairs multiple, 2-3 branched, lower head hairs single, distal pecten teeth usually with 1 (0-3) detached, pecten extended beyond the middle of siphon, siphon length 3 times width at base . . . . . *A. campestris*

2. Mesothoracic hair 1 long, 2 branched, hairs almost as long as head hairs 5 and 6, prothoracic hair formula 212-1-213, individual comb scales with median spine subequal to lateral spines, precratal area usually with 2-4 tufts, saddle extends 1/2-3/5 of the way down the sides . . . . . *A. dorsalis*

   Mesothoracic hair 1 very short, small, 2 branched, prothoracic hair formula 112-1-213, individual comb scales with prominent median spine, precratal area usually with 4-5 tufts, saddle extends 3/4 of the way down the sides . . . . *A. mediolineata*

First-instar larva

1. Siphon ratio (length/width at point of insertion of siphon hair) not greater than 2:1 . . . . . *A. dorsalis*

   Siphon ratio greater than 2:1 . . . . . (2)

2. Siphon hair reaching beyond apex of siphon . . . . . .
. . . . . *A. campestris*

* Siphon hair not reaching apex of siphon . . . . . .
. . . . . *A. mediolineata*

Female imago

1. Claws gradually curved, primary tooth 3 times as long as secondary tooth, the distance between primary and secondary tooth twice as long as the length of secondary tooth . . . .
. . . . . *A. dorsalis*

Claws abruptly curved, primary tooth twice as long as secondary tooth, the distance between primary and secondary tooth almost equal to the length of secondary tooth . . . . . (2)

2. Dark patches on abdominal tergites without white scales intermixed, broad median stripe on mesonotum spotted with white scales, wing scales predominantly dark especially on the distal part of the wing and costal vein . . . . . *A. mediolineata*

Dark patches on abdominal tergites scattered with white scales, segment 6 and 7 almost entirely white, broad median stripe on mesonotum entirely brown, wing scales predominantly white with dark scales intermixed . . . . . *A. campestris*

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* According to Dodge's (1966) description.

Male terminalia

1. Apical lobe of basistyle large, prominent, more or less quadrate, basal lobe with apex projecting upward, claspette filament longer than stem . . . . . *A. mediolineata*

   Apical lobe of basistyle round, small, inconspicuous . . . . .(2)

2. Basal lobe with large spines on the margin, one long recurved spine and one short stout spine, claspette filament swollen medially on convex side, filament shorter than stem, stem straight . . . . . *A. dorsalis*

   Basal lobe without large spines on the margin, claspette filament sickle-shaped, no prominent angle on convex side, filament as long as stem, stem curved . . . . . *A. campestris*

DISCUSSION

A. campestris, *A. dorsalis*, and *A. mediolineata* are three distinct species. The characters that separate the larvae and adults are listed in Table 9, and in the larval and adult keys.

The fourth-instar larvae of *A. campestris* can be separated from *A. dorsalis* and *A. mediolineata* by the number of branches on head hair 5. This hair is usually 2-3 branched in *A. campestris* and single in the other two species. Other reliable characters for separating *A. campestris* include the prothoracic hair formula, number of branches in mesothoracic hair 1, and the arrangement of the pecten teeth. These all differ in *A. campestris* and separate it from *A. dorsalis* and *A. mediolineata*. The difference in size of mesothoracic hair 1, the number of branches in prothoracic hair 1, the number of precratal tufts, and the shape of individual comb scales are the best characters for separating *A. dorsalis* from *A. mediolineata*.

The first-instar larvae of *A. campestris* can be separated from *A. dorsalis* most reliably by the siphonal ratio, which is greater in *A. campestris* than in *A. dorsalis*, and the length of the secondary sclerotized ring on the siphon, which is shorter in *A. dorsalis*. First-instar larvae of *A. mediolineata* (= Dodge, 1966 *A. melanimon*) is reported to have a shorter siphon hair than I have observed in *A. campestris*. If this is indeed true, then first-instar lavae of *A. mediolineata* can be separated from *A. campestris* as shown in the key. Similarly, the first-instar *A. dorsalis* is separated from *A. mediolineata* by the length of air tube and the length of the siphon hair.

With regard to the separation of adult males of *A. campestris*, *A. dorsalis*, and *A. mediolineata*, the shape of the apical and basal lobes, the setal arrangement of the basal lobe, the length of the claspette filament compared to the stem, shape of the claspette filament, and the size and shape of the metathoracic claws provided the most useful characters.

The most reliable character for separating females of *A. campestris* from the other two species is the appearance of white scales scattered on dark patches on the abdominal tergites. *A. campestris* can also be distinguished from *A. mediolineata* by the greater number of white scales on the costal wing vein in the former. *A. dorsalis* females can be separated from *A. campestris* reliably on the basis of the claw, and on the basis of the claw and the wing scales from *A. mediolineata*.

Most of the characters in *A. dorsalis* and *A. mediolineata* which I have examined agree with those studied by Richards (1956). One exception is the number of lateral hairs on abdominal segments 1 and 2 in larvae of *A. dorsalis* and *A. mediolineata*. Richards (1956) found 3 branches of lateral hairs on segments 1 and 2 in both species, while I found only 2 branches on both segments in these 2 species.

Mail (1934) and McLintock (1944) have pointed out that there are three different forms of *A. dorsalis*, each having a different type of mesonotal stripe. These were found in both the Montana and the Winnipeg populations by Mail and McLintock respectively. I have also observed these forms in a Saskatoon population. Each of these forms should be separated out and studied further by subsequent workers in mosquito biology and systematics.

Very little variation in adult females occurred in *A. campestris* and *A. mediolineata*. There was no significant difference in the

appearance of *A. campestris* larvae or adults collected from Churchill, Winnipeg or Saskatoon. Insufficient numbers of *A. mediolineata* from different locations did not permit detailed comparisons from different geographical regions.

The distribution of *A. campestris* in North America appears to be more northern than the other 2 species. It occurs in the western semi-arid plains and in the subarctic of North America whereas *A. mediolineata* is wide-spread in western North America. *A. dorsalis* has a holarctic distribution and was erroneously named a subspecies of *A. caspius* (Pallas) (Stackelberg, 1937). In the Palearctic region, *A. caspius* is represented by 2 forms (sub-species). The typical form, *A. caspius caspius*, occurs in more southern, arid regions with saline water. The other sub-species, *A. caspius dorsalis*, is characteristic of more northern and humid regions with fresh-water. *A. caspius dorsalis* (Monchadskii, 1951) from Europe is a synonym of *A. dorsalis* according to Stone *et al.* (1959). Natvig (1948) and Marshall (1938) considered *A. dorsalis* of Denmark and England respectively to be separate from *A. caspius*. Martini (1931) also considered *A. dorsalis* of Europe to be equivalent to *A. dorsalis* from North America.

In many places in the western United States, the three species appear to associate in the same area. In California, where *A. mediolineata* and *A. dorsalis* are widely distributed, *A. mediolineata* appears to be an inland, valley species and prefers fresh water compared to *A. dorsalis* which is primarily coastal and favors the margin of the lakes as well as bays for breeding places (Bohart, 1956).

Studies on voltinism in *A. campestris* from different locations indicated that this species is primarily multivoltine and could produce more than 1 generation per year. Part of the Churchill egg population

was found to be univoltine, and these eggs would require cold conditions before hatching would occur in nature. The results agreed with those of Chapman (1963) who studied *A. campestris* in Nevada and found that up to 95% of the eggs from some females hatched in the laboratory. These results indicate that *A. campestris* is actually a multivoltine species although it usually behaves as a univoltine species in the field. The suggestions for this behaviour and earlier observers' finding only 1 generation a year is probably due to the effect of ecological and climatological conditions. The oviposition sites have not been studied adequately enough to determine if they are flooded with summer rains, or if the site is only flooded during snow melt. In Winnipeg, *A. campestris* frequently occurs in such low numbers that it has been impossible to determine whether eggs are laid early enough in the summer to be ready to hatch with a summer rain. This species should be studied in areas where it is abundant each year, to determine the pattern of egg hatching under field conditions.

The adults of *A. campestris* are actively flying about by the middle to the end of June at Saskatoon, and by the middle of July at Churchill. In Winnipeg, they are not as numerous as *A. dorsalis*. Larvae appeared in the field as early as April 18 in roadside ditches with salinity on the alkaline side. Anal gills were very short (shorter than saddle on anal segment) in field-collected larvae at pH 8.1-8.4. Larvae reared in the laboratory at pH 7 had longer gills (3.6 times the saddle length). Although the larvae were normally found in the field at water temperatures of 12-30° C (McLintock, 1944), populations reared in the laboratory at Winnipeg were unable to complete larval development at a constant temperature of 15° C (Fig. 1). A temperature increase from 20° to 23° C reduced the

length of immature development by 5 and 7 days, in the Churchill and the Saskatoon population respectively. The lowest mortality occurred at 23°C, which indicates that this is the most suitable temperature, of the ones tested, for larval development.

Autogeny in *A. campestris* has been reported earlier from populations in Nevada by Chapman (1962). In the present study, autogeny was more common in a northern population than in a southern one. About 45% of the females of a Churchill population were capable of developing a few eggs autogenously, whereas in the Saskatoon population only 13.9% of the females developed eggs autogenously. This type of autogenous development in *A. campestris* is typically facultative autogeny (Corbet, 1967). Females began to develop a few follicles, usually after 10 days, and started laying eggs after 15-20 days. Typically autogenous species such as *Aedes rempeli*, *Aedes churchillensis*, *Aedes atropalpus* or *Wyeomyia smithii* begin egg development immediately after emergence and begin ovipositing within 4-8 days depending upon the species (Smith and Brust, 1970; Ellis and Brust, 1973; Brust, 1974; Evans and Brust, 1972).

The amount of blood had no effect on the number of eggs laid by females of *A. campestris* from Saskatoon and Estevan. The results from the Churchill population, showing a reduced fecundity, are quite difficult to evaluate, due to the influence of other factors such as age and the environmental conditions in the mating cage. The fecundity of the Churchill population can therefore not be compared to the other two populations. Two and three blood meals gave rise to more eggs than one blood meal in the Churchill population, but under the same conditions the other two populations may have reacted similarly.

Mating studies were all conducted in a large cage (120- x 120- x 210-cm). Further experiments should be conducted to attempt selection of a population that would mate in smaller cages, as was done with *A. dorsalis* (Blakeslee *et al.*, 1970; Grimstad *et al.*, 1970).

The results of short photoperiod (less than 14L:10D) indicate that a multivoltine population would enter diapause in the field during the latter part of August or early September. Univoltine eggs would require overwintering to terminate diapause under northern climates.

High temperature (30°C) and long photoperiod (16L:8D) terminated egg diapause in univoltine eggs of *A. campestris*. At 23°C and 16L:8D diapause was also terminated, but to a lesser degree than at 30°C.

SUMMARY

Samples of *A. campestris* from Churchill, Winnipeg, Saskatoon, and Estevan were collected and studied. These populations were primarily multivoltine with very few univoltine eggs except in the Churchill population. A successful laboratory colonization of *A. campestris* was maintained through 3 generations. About 20-30% mating was obtained in a 120-x 120-x 210-cm cage. The optimum temperature for development and survival of *A. campestris* from Churchill and Saskatoon in the laboratory was 23°C. The individuals reared at this temperature were larger than those reared at the other conditions. At 23°C, larval development takes 9 days and the pupal period is 3 days. Adult females complete their gonotrophic cycle within 3-4 days after blood feeding.

A. campestris is primarily anautogenous but autogeny is not uncommon since some females can complete their egg development without taking blood. The Churchill population exhibits 45% facultative autogeny, compared to 13.9% in the Saskatoon population. At 23°C, the number of blood meals had no effect on the fecundity of *A. campestris* from Saskatoon and Estevan.

Egg diapause in *A. campestris* can be induced by short photoperiod (<14L:10D) at 23° C. The critical photoperiod for inducing egg diapause in the Saskatoon population occurred between 13L:11D and 14L:10D. The percentage of diapausing eggs increased sharply after 10-14 days when exposed to a short photoperiod at 23°C. Long photoperiod (16L: 8D) terminated egg diapause in the Churchill population at 23°C after 7 days, and at 30°C the percentage termination was increased.

The anatomical features of first- and fourth-instar larvae and adults of *A. campestris*, *A. dorsalis*, and *A. mediolineata* are illustrated and tabulated. Keys to all stages are included.

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