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The comparative morphology of antennal structures in Buprestidae (Coleoptera): evolutionary trends, taxonomic and phylogenetic implications. Part 1.

MARK G. VOLKOVITSH

Zoological Institute, Russian Academy of Sciences 199034 St.Petersburg, Russia; e-mail: polycest@zin.ru

VOLKOVITSH M. G. 2001: The comparative morphology of antennal structures in Buprestidae (Coleoptera): evolutionary trends, taxonomic and phylogenetic implications. Part 1. *Acta Musei Moraviae, Scientiae biologicae* (Brno) **86:** 43–169. – The results of comparative morphological study of antennal structures in Buprestidae LEACH, 1815 are presented and illustrated with scanning electron microphotographs. In total, the antennae of 412 species from 316 genera of all the subfamilies of Buprestidae and 6 genera of other Elateriformia were studied using scanning electron microscopy. In addition, the antennae of many species from other buprestid genera were examined using optic dissecting and compound microscopes. The general structures of antennae, individual sensillae and sensory organs in Buprestidae are described with particular attention given to the structure and arrangement of antennal sensory organs as key taxonomic characters. It is established that the structure of entire antennae and antennomeres separately, the structure, composition, placing, and quantitative characteristics of antennal sensillae and sensory organs, as well as their sexual dimorphism, may serve as reliable taxonomic characters. A review of antennal structures in subfamilies Schizopodinae LE CONTE, 1861, Julodinae LACORDAIRE, 1857, Polycestinae LACORDAIRE, 1857, Chalcophorinae LACORDAIRE, 1857, Buprestinae, and Galbellinae REITTER, 1911 is followed by a discussion of their genesis, evolutionary trends, and taxonomical and phylogenetic implications. Each main phyletic stock (complex) of Buprestidae is characterised by certain evolutionary trends and particular pathways of genesis of antennal structures. The smaller phyletic lineages demonstrate their own morphoclines although the separate states can appear to be quite similar in the representatives of different lineages or even stocks. The most primitive states are found in Schizopodinae and Julodinae, although the latter demonstrate a rather high level of specialisation. Polycestinae and Buprestinae show quite similar (buprestoid) pathways of antennal structure genesis, whereas the taxa attributed to the Chalcophoroid complex exibit a particular (chalcophoroid) pathway. The evolutionary trends of antennal structures in Galbellinae are somewhat unclear. Based on peculiarities of antennal structure genesis, the main phylogenetic stocks, lineages, branches and groups are recognised in Buprestidae. The systematic position, taxonomic status and relationships of many other buprestid higher taxa are discussed. Taxonomical changes and suggestions are listed in the Summary. A list is provided of the buprestid taxa examined using a scanning electron microscope, arranged with respect to their antennal structure.

Key words: Coleoptera, Buprestidae, antennal structures, sensillae, sensory organs, evolution, taxonomic and phylogenetic implications

Introduction *)

The antennal structures of the Buprestidae, i.e. the shape and number of antennomeres, and particularly the arrangement of what are known as the sensory or

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sensitive pores, pits, fovea or fossae, are widely used for the systematics and classification of the group, although some researchers have been critical of the taxonomical value of these characters. Antennal sensory organs are of considerable current use in insect systematics. For example, a revised classification of the tenebrionid beetles (MEDVEDEV 1977) has been based on these structures. Almost nothing has been published on antennal sensory formations in the Buprestidae with the exception of a description of highly specialised antennal organs in two *Melanophila* species (SCOTT & GARRA 1975), and a short review of antennal structure in Stigmoderini (GARDNER 1989); both publications were illustrated with scanning electron micrographs. An attempt to evaluate and apply antennal characters to buprestid systematics was recently made by VOLKOVITSH (1990). New data on the structure of buprestid antennae resulted from extensive additional study, and their application to buprestid taxonomy, phylogeny and classification is presented below.

The tribal delimitation of the Buprestidae by LACORDAIRE (1857), which was largely based on antennal characters, lies at the heart of many subsequent classifications (MARSEUL 1865, 1866, LECONTE & HORN 1883, KERREMANS 1902, 1903, OBENBERGER 1926, 1930, 1934, 1935, 1936, 1937, RICHTER 1949, 1952, COBOS 1955, 1975a, 1979a,b, 1980, 1981, 1986a, and others). According to this traditional concept, given general currency by BELLAMY (1985), the Buprestidae is made up of 14 subfamilies, 51 tribes and 428 genera; since then Schizopodinae has been raised to family rank (NELSON & BELLAMY 1991), some new tribes have also been constructed and tens of new genera described. A different and non-traditional concept of buprestid classification was recently presented by HOŁYNSKI (1988, 1993). He held that the family is made up of only 4 subfamilies (Schizopodinae, Julodinae, Buprestinae, and Agrilinae), 12 tribes, and 65 subtribes. HOŁYNSKI (1988, 1993) downgraded many traditional tribes to subtribe level and significantly changed their generic composition. He also proposed a number of new subtribes. Both of the above-mentioned classifications have the essential drawback that they lack a reliable morphological foundation, resulting in the use of casual, contradictory, and variable characters reflecting general similarity rather than real phylogenetic relations.

The main objectives of this work are as follows: the detailed examination, comparison and evaluation of the taxonomical value of buprestid antennal structures at the level of the higher taxa, with particular attention given to the structure and arrangement of sensory organs as key taxonomical characters; the establishment of morphoclines of their transformations and general evolutionary trends; the clarification of possible relationships of taxa that have similar states, in order to clear up their phylogeny and refine their taxonomical positions. *It is vital to note that I do not suggest a new classification of the Buprestidae, because in my opinion the creation of a natural classification based on a single character system is impossible.* The order in which the taxa are presented here largely follows the traditional classification (BELLAMY 1985) with all remarks and suggestions regarding taxonomical changes discussed in the final sections of this paper.

Material and methods

Most of the materials for this research came from the collection of the Zoological Institute [ZIN], Russian Academy of Sciences, Saint Petersburg (Russia). Some additional material was loaned by the National Museum, Prague (Czech Republic), the National Museum of Natural History, Smithsonian Institution, Washington, D.C. (U.S.A.), the Natural History Museum, Budapest (Hungary), the Transvaal Museum, Pretoria (South Africa), and the collections of Dr. C. L. Bellamy, Pretoria (South Africa) and V. Kubáň, Brno (Czech Republic).

Antennae were examined using the Jeol scanning electron microscope at the Institute of Biology of Inner Waters [IBIW], the Russian Academy of Sciences, Borok (Yaroslavl region, Russia) in 1984–1987, and a Hitachi SEM at the Zoological Institute, the Russian Academy of Sciences, Saint Petersburg (Russia) in 1988–1996. Separated antennae were covered with a gold or platinum monolayer and were studied under magnifications from ×35 to ×15,000. A total of 412 species from 316 genera (approximately 67% of known genera), belonging to nearly all the tribes (excepting Perucolini) of all the buprestid subfamilies, as well as 6 genera from other families among the Elateriformia (Dascilloidea: Dascillidae; Byrrhoidea: Byrrhidae, Dryopidae, Heteroceridae; Elateroidea: Elateridae, Lycidae) (Lawrence & Newton 1995) were studied and 1,861 micrographs were made in the course of the investigation. In addition, a great number of species from several genera were examined using an MBS-9 optical dissecting microscope.

Material examined

Aaata Sem. [*A. finchi* (Waterh.)], *Achardella* Obenb. [*A. americana* (Hbst.)], *Acherusia* (s.str.) Lap. & Gory [*A. childreni* Lap. & Gory], *Acmaeodera* (*Acmaeotethya* Volk.) [*A. pallidepicta* Reitt.], *Acmaeodera* (*Cobosiella* Volk.) [*A. chotanica* Sem.], *Acmaeodera* (*Palaeotethya* Volk.) [*A. bipunctata* (Ol.)], *Acmaeodera* (*Paracmaeodera* Théry) [*A. elevata* (Klug)], *Acmaeodera* (*Ptychomus* Mars.) [*A. polita* (Klug)], *Acmaeodera* (*Rugacmaeodera* Holm) [*A. subprasina* Mars.], *Acmaeodera* (s.str.) Eschsch. [*A. rubronotata* Lap. & Gory, *A. babatauensis* Obenb., *A. chalcithorax* Obenb.], *Acmaeoderella* (*Carininota* Volk.) [*A. flavofasciata* (Pill. & Mitt.)], *Acmaeoderella* (*Euacmaeoderella* Volk.) [*A. staudingeri* (Ab.), *A. gibbulosa* (Mén.), *A. villosula* (Stev.), *A. subcyanea* (Reitt.), *A. solskyi* (Obenb.)], *Acmaeoderella* (s.str.) Cobos [*A. caspica* (Ganglb.)], *Acmaeoderoides* Van Dyke [*A. knulli* Nels.], *Acmaeoderopsis* Barr [*A. jaguarina* (Knull)], *Actenodes* Dej. [*A. costipennis* Lap. & Gory], *Afrobothris* Théry [*A. nigrita* (Ol.)], *Agaeocera* Saund. [*A. scintillans* Waterh.], , *Agelia* Lap. & Gory [*A. peteli* (Gory)], *Aglaostola* Saund. [*A. tereticollis* (Pall.)], *Amblysterna* Saund. [*A. natalensis* (Fåhr.)], *Ampheremus* Fall [*A. cylindricollis* Fall.], *Anambodera* Barr [*A. palmarum* (Timb.)], *Anilara* Saund. [*A. sulcicollis* Kerr.], *Anthaxia* (*Haplanthaxia* Reitt.) [*A.* sp.], *Anthaxia* (*Melanthaxia* Richter) [*A. strigiata* Le Conte, *A. aeneogastra* Horn, *A. inornata* Rand., *A.* sp.], *Anthaxia* Eschsch. (s.str.) [*A. vittula* Kiesw.], *Anthaxoschema* Obenb. [*A. terraereginae* Obenb.], *Aristosoma* Saund. [*A. suturale* (Thunb.)], *Armenosoma* Waterh. [*A. atrum* Waterh.], *Asamia* Théry [*A. insolita* Théry], *Asidoptera* Obenb. [*A. monstruosa* Kerr.], *Astraeus* Lap. & Gory [*A. flavopictus* Lap. & Gory, *A. mastersi* Macl.], *Augrabies* Bell. [*A. schotiaphaga* Bell.], *Balthasarella* Obenb. [*B. melandryoides* Obenb.], *Baudonisia* Cobos [*B. villosiventris* (Chevr.)], *Beerellus* Nels. [*B. taxodii* Nels.], *Belionota* Eschsch. (s.str.) [*B. aenea* Deyr., *B. sumptuosa* Lap. & Gory], *Bellamyina* Bílý [*B. hunanensis* Peng], *Brachanthaxia* Théry [*B. gemmata* (Lap. & Gory)], *Brachelytrium* Obenb. [*B. transvaalense* Obenb.], *Bubastes* Lap. & Gory [*B. iridescens* Théry, *B. inconsistans* Thoms.], *Bulis* Lap. & Gory [*B. bivittata* (F.)], *Buprestina* Obenb. [*B. prosternalis* Obenb.], *Buprestis* (*Orthocheira* Richter) [*B. salomoni* Thoms.], *Buprestis* L. (s.str.) [*B. strigosa* Gebl.], *Callistroma* Fairm. [*C. oxypyra* Fairm.], *Callopistus* Deyr. [*C. castelnaui* Deyr.], *Calodema* Lap. & Gory [*C. regalis* Lap. & Gory, *C. kirbyi* (Hope)], *Capnodis* Eschsch. [*C. tenebrionis* (L.), *C. parumstriata*

Ball.], *Cardiaspis* Saund. [*C. mouhotii* Saund.], *Catoxantha* Dej. [*C. opulenta* Gory], *Chalcogenia* Saund. [*C. contempta* (Klug)], *Chalcophora* Dej. [*C. mariana* (L.), *C. japonica* (Gory)], *Chalcophorella* Kerr. [*C. stigmatica* (Schoenh.)], *Chalcophoropsis* Saund. [*C. quadrifoveolata* (Lap. & Gory)], *Chalcoplia* Saund. [*C. auripilis* Obenb.], *Chalcopoecila* Saund. [*C. ornata* (Gory)], *Chlorophorella* Desc. [*C. gerlingi* Desc.], *Chrysaspina* Théry [*C. aurata* (F.)], *Chrysesthes* Sol. [*C. gymnopleura* Perty], *Chrysobothris* (*Abothris* Sem. & Richter) [*C. nana* Fairm.], *Chrysobothris* Eschsch. (s.str.) [*C. affinis* (F.), *C. chrysostigma* (L.), *C. dentipes* Lap. & Gory], *Chrysodema* Lap. & Gory [*C. occulta* Waterh.], *Chrysophana* Le Conte [*C. placida* Le Conte], *Cinyra* Lap. & Gory [*C. obenbergeri* (Cobos)], *Colobogaster* Sol. [*C. biguttatus* Kerr.], *Conognatha* Eschsch. [*C. pretiosissima* Chevr.], *Coomaniella* Bourg. [*C. taiwanensis* Baud., *C. biformis* Bílý & Kalashian], *Cordillerita* Obenb. [*C. bruchi* Obenb.], *Cratomerus* (*Cryptocratomerus* Richter) [*C. iliensis* (Obenb.)], *Cratomerus* Sol. (s.str.) [*C. hungaricus* (Scop.)], *Cromophila* Cobos [*C.* sp.], *Curis* Lap. & Gory [*C. caloptera* (Boisd.)], *Cyalithus* Thoms. [*C. rugifrons* (Deyr.)], *Cylindrophora* Sol. [*C. maulica* (Mol.), *C. verecunda* (Erich.)], *Cyphogastra* Deyr. [*C. foveicollis* (Boisd.)], *Cyphogastrella* Théry [*C. gratiosissima* (Kerr.)], *Cyphosoma* Mnnh. [*C. tataricum* (Pall.)], *Cyria* Sol. [*C. imperialis* (F.)], *Dactylozodes* Chevr. [*D. confus* Fairm.], *Descarpentriesiola* Cobos [*D. freyi* (Théry)], *Diadoxus* Thoms. [*D. erythrurus* White, *D. scalaris* Lap. & Gory], *Dicerca* (*Argante* Gistel) [*D. moesta* (F.)], *Dicerca* Eschsch. (s.str.) [*D. furcata* (Thunb.), *D. aenea* (L.)], *Dicercomorpha* Deyr. [*D. albosparsa* (Lap. & Gory)], *Ditriaena* Waterh. [*D. sexspinosa* (Waterh.)], *Dystaxia* Le Conte [*D. elegans* Fall], *Ectinogonia* Spin. [*E. buqueti speciosa* (Germ.)], *Embrikillium* Obenb. [*E. mirandum* Obenb.], *Epidelus* Deyr. [*E. wallacei* (Thoms.)], *Epistomentis* Sol. [*E. pictus* (Gory)], *Eububastes* Obenb. [*E. nickerli* Obenb.], *Euchroma* Sol. [*E. gigantea* (L.)], *Euleptodema* Obenb. [*E. nigrum* (Thoms.)], *Euplectalecia* Obenb. [*E. erythropus* (Gory), *E. pulverulenta* (Waterh.)], *Eupodalecia* Obenb. [*E. minarum* Obenb.], *Euryspilus* Lac. [*E.* sp.], *Eurythyrea* Dej. [*E. aurata* (Pall.)], *Evagora* Kerr. [*E. amorpha* Obenb.], *Evides* Serv. in Dej. [*E. kraatzi* Kerr., *E. gambiensis* (Lap. & Gory)], *Exagistus* Deyr. [*E. igneiceps* Deyr.], *Fahraeusia* Obenb. [*F. chalcea* Obenb.], *Galbella* Westw. [*G. felix* (Mars.), *G.* sp.], *Gelaeus* Waterh. [*G. walkeri* Waterh.], *Genestia* Théry [*G. semenovi* Obenb.], Genus 1 [sp. 1], *Glyptoscelimorpha* (*Distaxiella* Knull) [*G. juniperae* Knull], *Glyptoscelimorpha* (s.str.) Horn [*G. marmorata* Horn, *G. viridis* (Chamb.)], *Halecia* Lap. & Gory [*H. blanda* (F.)], *Haplotrinchus* Kerr. [*H. inaequalis* (Deyr.)], *Hesperorhipis* Fall [*H. jacumbae* Knull], *Hilarotes* Saund. [*H. nitidicollis* (Lap. & Gory)], *Hiperantha* (*Hyperantella* Hosch.) [*H. interrogationis* (Klug)], *Hiperantha* Gistel (s.str.) [*H. haemorrhoa* Fairm.], *Hippomelas* Lap. & Gory (s.str.) [*H. planicauda* Casey], *Hypoprasis* Fairm. & Germ. [*H. harpagon* Fairm. & Germ.], *Hypostigmodera* Blackb. [*H. variegata* Blackb.], *Iridotaenia* Deyr. (*Iridomroczkowskia* Hoł.) [*I. sulcata* (Thunb.)], *Jakovleviola* Obenb. [*J. strandi* Obenb.], *Jelinekia* Cobos [*J. barri* (Nels.)], *Julodella* Sem. [*J. kaufmanni* (Ball.)], *Julodimorpha* Gemm. & Har. [*J. bakewelli* White], *Julodis* Eschsch. [*J. euphratica* Lap. & Gory, *J. variolaris* (Pall.)], *Juniperella* Knull [*J. mirabilis* Knull], *Kisanthobia* Mars. [*K. ariasi* (Robert)], *Kurosawaia* Toyama & Ohmomo [*K. yanoi* (Kuros.)], *Lampetis* Spin. [*L. argentata* (Mnnh.)], *Lamprocheila* Saund. [*L. maillei* (Lap. & Gory)], *Latipalpis* (*Palpilatis* Bílý) [*L. plasoni* (Reitt.)], *Latipalpis* Sol. (s.str.) [*L. plana* (Ol.)], *Maoraxia* Obenb. [*M. eremita* (White), *M. littoralis* Bell. & Will., *M. excavata* (Fauv.)], *Mastogenius* Sol. [*M. cyaneus* Fish., *M. robustus* Schaeff.], *Melanophila* Eschsch. (s.str.) [*M. acuminata* (DeG.)], *Melobasis* (*Briseis* Saund.) [*M. tenebrosa* Obenb.], *Melobasis* Lap. & Gory (s.str.) [*M. gloriosa* Hope, *M.* sp.], *Mendizabalia* Cobos [*M. germaini* (Germ. & Kerr.)], *Merimna* Saund. [*M. atrata* (Hope)], *Metataenia* Théry (*Metamroczkowskia* Hoł.) [*M. clotildae* Gestro], *Micrasta* Kerr. [*M. creola* Obenb.], *Microacmaeodera* (s.str.) Cobos [*M. longicornis* (Cobos)], *Microcastalia* Hell. [*M. globithorax* (Thoms.)], *Micropistus* Théry [*M. microcephalus* Théry], *Mixochlorus* Waterh. [*M. lateralis* Waterh.], *Montrouzieretta* Obenb. [*M. caledonica* Obenb.], *Nanularia* Casey [*N. brunneata* (Knull)], *Nascio* Lap. & Gory [*N. vetusta* (Boisd.)], *Nascioides* Kerr. [*N. enysii* (Sharp.)], *Neobubastes* Blackb. [*N. australiasiae* Obenb.], *Neobuprestis* Kerr. [*N. australis* (Blackb.)], *Neocuris* Saund. [*N.* sp. 1, *N.* sp. 2], *Neocuropsis* Obenb. [*N. splendens* Macl.], *Neocypetes* Cobos [*N. guttulata* (Fairm. & Germ.)], *Neojulodis* Kerr. (*Protojulodis* Holm) [*N. vittipennis* (Fåhr.)], *Neotorresita* Obenb. [*N. achardi* Obenb.], *Nesotrinchus* Obenb. [*N. orientalis* Bell.], *Nipponobuprestis* Obenb. [*N. amabilis* (Snell.van Vollenh.)], *Nothomorpha* Saund. [*N. maior* Kerr.], *Nothomorphoides* Holm [*N. irishi* Holm], *Notographus* Thoms. [*N. sulcipennis* (Macl.)], *Odettea* Baud. [*O. laosensis* Baud.], *Oedisterna* Lac. [*O. cuprea* (L.)], *Palmar* (*Scintillatrix* Obenb.) [*P. chinganensis* (Obenb.), *P. limbata* (Gebl.)], *Palmar* Schaefer (s.str.) [*P. virgata* (Motsch.)], *Papuodema* Obenb. [*P. quadristigmosa* Obenb.], *Paracastalia* Kerr. [*P. ornatipennis* Kerr.], *Paracupta* Deyr. [*P. xanthocera* (Boisd.)], *Paracuris* Obenb. [*P. bimaculata* (Gory)], *Paraphrixia*

Saund. [*P. purpurea* (Ol.)], *Parataenia* Kerr. [*P. orbicularis* Kerr.], *Paratassa* Mars. [*P. coraebiformis* (Fairm.)], *Paratrachys* Saund. [*P. hederae* Saund.], *Paraxenopsis* Cobos [*P.* sp.], *Pelecopselaphus* Sol. [*P. acutus* Saund.], *Peronaemis* Waterh. [*P. monticola* Fish.], *Perotis* Dej. [*P. lugubris* (F.)], *Phaenops* Dej. [*P. guttulata* (Gebl.)], *Philandia* Kerr. [*P. valdiviana* (Phil. & Phil.)], *Philanthaxia* Deyr. [*P.* sp.], *Philocteanus* Deyr. [*P. plutus* (Lap. & Gory)], *Phrixia* Deyr. [*P. albomaculata* Fish., *P.* sp.], *Poecilonota* Eschsch. [*P. variolosa* (Payk.)], *Polybothris* Dup. [*P. sulcicollis* Kerr., *P. staudingeri* Théry], *Polycesta* Dej. [*P. porcata* (F.)], *Polycestina* (*Damarana* Cobos) [*P. damarana* (Kerr.)], *Polycestina* (s.str.) Cobos [*P. quatuordecimmaculata* (Fåhr.)], *Polyctesis* Mars. [*P. rhois* Mars.], *Prospheres* Thoms. [*P. aurantiopictus* (Lap. & Gory), *P. chrysocomus* Fauv.], *Pseudacherusia* Kerr. [*P. bartoni* Obenb.], *Pseudactenodes* Kerr. [*P. vitticollis* (Har.)], *Pseudanilara* Théry [*P. cupripes* (Macl.)], *Pseudocastalia* Kraatz [*P. arabica* Gestro], *Pseudolampetis* Obenb. [*P. circumsulcata* (Lap. & Gory)], *Pseudoperotis* Obenb. [*P.* sp.], *Pseudotaenia* Kerr. [*P. salamandra* (Thoms.)], *Pterobothris* Fairm. & Germ. [*P. corrosus* Fairm. & Germ.], *Ptosima* Dej. [*P. undecimmaculata* (F.), *P. gibbicollis* (Say)], *Pygicera* Kerr. [*P. scripta* (Lap. & Gory)], *Pygichaeta* Obenb. [*P. semigranosa* (Sol.)], *Rhabdolona* Obenb. [*R. strandi* Obenb.], *Sapaia* Bílý [*S. brodskyi* Bílý], *Saundersina* Cobos [*S. modesta* (F.)], *Scaptelytra* Saund. [*S. oculicollis* Kerr.], *Schizopus* Le Conte [*S. laetus* Le Conte], *Schoutendeiastes* Bourg. [*S. amabilis* (Lap. & Gory), *S. ohkurai* (Akiyama & Ohm.)], *Spectralia* Casey [*S. purpurascens* (Schaeff.)], *Sphenoptera* (*Chrysoblemma* Jak.) [*S. beckeri* Dohrn, *S. potanini* Jak.], *Sphenoptera* (*Sphenopterella* Volk. & Kalash.) [*S. margaritae* Volk. & Kalash.], *Sphenoptera* Dej. (s.str.) [*S. glabrata* (Mén.)], *Sponsor* (s.str.) Lap. & Gory [*S. emmae* Desc., *S. kerremansi* Théry], *Steraspis* Dej. [*S. squammosa* (Klug)], *Sternocera* Eschsch. [*S. orissa luctifera* (Klug)], *Stigmatophorella* Obenb. [*S. quadrioculata* (Redt.)], *Stigmodera* Eschsch. [*S. macularia* (Donov.)], *Strigoptera* Dej. [*S. bimaculata* (L.)], *Strigopteroides* Cobos [*S. aegyptiacus* (Gmel.)], *Tamamushia* Miwa & Chujô [*T. virida* Miwa & Chujô], *Tetragonoschema* Thoms. [*T.* sp.], *Texania* Casey [*T. campestris* Say], *Themognatha* Sol. [*T. variabilis* (Donov.)], *Theryaxia* Cart. [*T. suttoni* Cart.], *Thomassetia* Théry [*T. crassa* (Waterh.)], *Thrincopyge* Le Conte [*T. alacris* Le Conte, *T. ambiens* (Le Conte), *T. marginata* Waterh.], *Thurntaxisia* Schatzm. [*T. alexandri* Schatzm., *Thymedes* Waterh. [*T. flavicornis* (Saund.)], *Torresita* Gemm. & Har. [*T. dilatata* (Redt.)], *Touzalinia* Théry [*T. psilopteroides* Théry], *Trachykele* Mars. [*T. blondeli* Mars.], *Trachypteris* Kirby [*T. picta* (Pall.)], *Trichinorhipis* Barr [*T. knulli* Barr], *Trigonogenium* Har. [*T. angulosum* (Sol.)], *Tylauchenia* Burm. [*T. crassicollis* (Lap. & Gory)], *Tyndaris* (*Paratyndaris* Fish.) [*T. olneyae* Skin.], *Tyndaris* (s.str.) Thoms. [*T. planata* (Lap. & Gory)], *Vadonaxia* Desc. [*V. peyrierasi* Desc.], *Xantheremia* (s.str.) Volk. [*X. subscalaris* (Reitt).], *Xenocyria* Obenb. [*X. vittigera* (Lap. & Gory)], *Xenomelanophila* Sloop [*X. miranda* (Le Conte)], *Xenorhipis* Le Conte [*X. brendeli* Le Conte], *Xyroscelis* Thoms. [*X. crocata* (Lap. & Gory)], *Yamina* Kerr. [*Y. sanguinea* (F.)],

Results

I. The general structure of antennae and sensory formations in Buprestidae

Antennae

Buprestid antennae are usually serrate or serrate-truncate, rarely bead-like, pectinate, flabellate, geniculate or anomalous in shape; generally comprising 11, very seldom 10 or 12, antennomeres. The first (scape) and the second (pedicle) proximal antennomeres are basically invariable in all the taxa, bearing only trichoid sensillae superficially. Antennomeres 3–11(12) (flagellum) are highly variable in shape and size within the same genera, species groups or even between the sexes, being more or less symmetrical or asymmetrical, bilaterally and/or transversely flattened, transversely expanded or elongated, attaching perpendicularly or at an angle to longitudinal or transverse antennal axes. The apical antennomere usually differs in shape and sensory organ location.

To describe the arrangement of sensory formations I consider the antennomere side facing the body when the antennae are in repose as the 'internal' and the opposite side as the 'external'; the proximal part of the antennomere as its base and the distal one as its apex; the margin ajacent to the longitudinal antennal axis as the outer, its opposite as the inner, and the apical margin as anterior (when the antennomere is swollen it has an anterior surface often bearing the sensory organs); the corner between anterior and inner margins, often extending into a projection, is an 'apical corner'.

Visible sensory formations vary greatly in shape, sensillary composition and structure. They appear to begin from antennomeres 4 or 5, only rarely from 3, 6, or even 7–9, occupying the internal side. These structures may occur on both internal and external sides, sometimes dispersing all over the surface, or they may be located only on the anterior surface. Sensory formations are most abundant on the apical antennomere, often being well developed on both sides.

In many taxa the antennae exhibit sexual dimorphism, in which the male antennomeres are substantially expanded (Fig. 17) or transformed (Fig. 148). This is frequently accompanied by the presence of modified, presumably olfactory sensillae (Figs 11, 148, 192, 194, 211, 218) never occurring in females, or a predominance of usual basiconic sensillae. It is important that when the males have strongly modified antennae, the females retain the sensory formations typical of certain group (Figs 18, 149).

The correlation between antennomere shape and sensory formation type may be easily observed. As a rule, bilaterally flattened triangular antennomeres bear large apical or subapical fossae (Figs 21, 88). Serrate-truncate or elongated rectangular antennomeres bear a small apical pit or cavity (Figs 42, 132, 133), extensive sensillary fields (Figs 5, 40, 43, 90, 123), or numerous pits dispersed over the whole surface (Figs 56–65), or both apical and lateral fossa internally and lateral field or fossae externally (Figs 80–81, 99, 100). Transversely flattened antennomeres have a big fossa on the anterior surface (Figs 1, 46, 92, 113), often with sensillary fields as well (Figs 45, 47, 93, 141); flabellate or pectinate antennomeres usually have only individual sensillae (Figs 116, 148), widely dispersed sensory pores and pits (Figs 52–53, 73) or poorly developed apical organs (Fig. 118), etc.

The examination of buprestid antennae using a scanning electron microscope has shown that what have been known as antennal chaetae, pores, pits and fossae prove to be individual sensillae of different types and their clusters, attached to the surface or deepened into the cuticle to form compound sensory organs. As a rule, each antennomere bears both individual sensillae and sensory organs.

Sensillae

The term sensilla is used here to mean only the terminal cuticular part of a sensory organ; an examination of ultrastructure and function of sensillae was not involved in this research, so the identification and classification of sensillae has been compiled from ZACHARUK (1985) and ALM & HALL (1986). The general types of buprestid antennal sensillae are classified as chaetoid, trichoid, squamoid, basiconic, and campaniform.

There are also strongly modified sensillae that are found exclusively in males. The mode of attachment to the cuticle is also significant, with the following types of sensillae recognised: superficial, coeloconic (deepened into cuticle depressions), and styloconic (arising from cuticular tubercles). Some of the pores occurring on the antennomere surface (Figs 33, 117) could be considered as terminal openings of scolopophores or ampullar sensillae, but the wax particles usually surrounding these pores suggest that they are in all probability the apertures of wax glands. None of the placoid sensillae that occur in many coleopteran groups (e.g. in some Tenebrionidae) (MEDVEDEV 1977) were found in the Buprestidae; among other Elateriformia under study, such a sensillary type was found exclusively in Lycidae. Sensillae are receptors with different functions (e.g. mechanoreceptors, hygroreceptors, chemoreceptors, thermoreceptors, gustatory and olfactory receptors, etc.), but as is currently accepted, many sensillae are multifunctional organs.

The main types of buprestid sensillae are as follows:

♦ Type U1. Uniporous trichoid and chaetoid groove-walled sensillae (Figs 2–3, 10, 31, 33, 65, 107, 117, 159, U1) are long, round in cross-section and blunted. They bear a single pore apically and are arranged singly or in small groups on apical corners and near the anterior margins. In rare cases they occur on the sides or along the inner margins (Fig. 65). These sensillae are presumably contact chemoreceptors (gustatory).

♦ Type U2. Uniporous basiconic groove-walled superficial, coeloconic or styloconic sensillae (Figs 94–95, 107, U2), apparently derived from the previous type, are short, peg- or cone-like, round in cross-section, blunted and bearing a single pore apically. They are arranged singly or in small groups, sometimes forming extensive fields on the sides and along the inner margin (Figs 94–95, 102–103, 107, 113, 152–155). These sensillae are also presumably contact chemoreceptors.

♦ Type A3. Aporous trichoid, chaetoid, and squamoid groove-walled sensillae (Fig. 2, 8–10, 21, 31, 33–34, 107, 117, A3) are long, flattened in cross-section, sharp or serrated apically. They have no pores, and are arranged singly or in groups more or less evenly dispersed or, rarely, forming extensive fields (Figs 5–6, 8–9). These sensillae mainly occupy the sides and outer margins of antennomeres and are most abundant on proximal segments. They presumably serve as mechanoreceptors and chemoreceptors.

♦ Type B4. This type contains multiporous basiconic smooth-walled superficial, styloconic, and coeloconic sensillae. They presumably serve as chemoreceptors (olfactory sensillae), and perhaps as thermo- or hygroreceptors as well. These sensillae may be distinguished as follows:

•Subtype B4a. Peg-like or cone-like sensillae with filamentous apices (Figs 3, 7, 34, B4a; 151, 157) are usually incorporated into apical organs.

•Subtype B4b. Peg-like or cone-like sensillae with rounded apices (Figs 3, 7, 31, B4b) are also incorporated into the apical organs; they may be combined with sensillae of the previous subtype.

•Subtype B4c. Cone-like sensillae (Figs 2–3, 8–9, 21, 31, 33, 117, 159, B4c) differ from the others in occurring mainly separate from sensory organs and often forming extensive fields on the sides and along inner margins (Figs 9, 32–33, 38, 40, 43, 121–134). Only rarely are they concentrated around sensory organs or incorporated into them (Figs 13, 15–18, 19–20, 22, 31).

♦Type M5. Multiporous basiconic groove-walled sensillae (Figs 3, 117, M5) differ from the others in having a costate apical part and a smooth base. They are usually incorporated into apical organs and occur singly or in very small groups. These sensillae are presumably distant chemoreceptors.

♦Type M6. Multiporous basiconic sensillae with digitiform apices found only in certain genera. They resemble the previous type and apparently share derivation with them, performing the same function but having smooth walls. These sensillae are located inside apical organs.

♦Type C7. Campaniform sensillae (Figs 3, 117, C7) are incorporated into apical organs and arranged in small groups, presumably as chemoreceptors.

♦Type S8. Heterogenous group including specialised male sensillae (Figs 117, S8; 192, 194, 211; 219, ?S8) that occur in some taxa from practically all the subfamilies, often in a single species, or a few species from the same genus or species group. They are extremely variable in shape and size, usually forming extensive fields or dispersed over the surface of the antennomere (Figs 11, 14, 17, 116, 118, 148). These sensillae are presumably distant (olfactory) chemoreceptors perceiving the sex pheromones of females.

The above-mentioned types do not cover the whole diverse range of buprestid sensillae. The antennae of some groups carry sensillae that cannot be assigned to any of the types listed. The catoxanthoid sensillae described earlier in Chrysochroini as a distinct type (VOLKOVITSH 1990) proved to be an artifact consisting of the bases of broken basiconic sensillae. With regard to position, two basic groups of antennal sensillae might be recognised. Group 1 consists of external sensillae dispersed singly or in small groups (U1, A3, S8), or forming superficial sensillary fields (U2, B4c, rarely A3). Group 2 consists of internal sensillae incorporated into sensory organs (B4a, B4b, M5, M6, C7). Generally, there are no rigid boundaries between sensillary types and subtypes; for example, the sensillae of B4a and B4c subtypes often transform into a B4b subtype when concentrated in deep depressions.

Buprestid taxa differ in having different patterns, composition, and quantitative characters of antennal sensillae, the manner of their attachment to the cuticle (superficial, coeloconic or styloconic), and placing of which may serve as diagnostic characters. Significant distinctions in sensillary characters may be found even within the same genera or species groups (e.g. within Xenorhipini, Figs 191–195), although the general type of sensillary structures is usually fixed.

Sensory organs

Sensory organs that are aggregations of sensillae and cuticle structures may be distinguished as follows:

1) distinct sensory organs absent, individual sensillae are more or less uniformly dispersed over antennomere surface (Figs 116, 148).

2) sensillary groups consisting of several individual sensillae of the same type (Figs 54, 77, 92, 135–136; U1 sensillae on anterior corners and on top of apical antennomere).

3) sensory fields that are extensive areas bearing numerous superficial (Figs 5, 9, 33, 38, 43, 121–131, 144, 153–155, 180, 198–201, 205) or coeloconic (Figs 18, 40, 62, 90–94, 141, 206) sensillae of one or more types, or sensillary pores and small pits usually located on the sides and along the inner margins of antennomeres (Figs 45–50, 52–56, 63–65, 73–74; 95, LF).

4) sensory zones that are small, superficial, undefined or poorly defined, sometimes elevated sensillary areas (Figs 6, SZ; 7, 10, 13, 19–20, 34).

5) sensory depressions that are poorly defined, shallowly sloping into the cuticle, usually irregular in shape, having no distinct boundaries to the sensillary areas (Figs 1, AD; 2–3, 26, 41, 45–46, 58, 79; 95, AD; 118, SD; 136, 142, 217, 219–220).

6) sensory pits and fossae that are well defined, round, deeply sloping into cuticle, usually regular, having distinct boundaries to the sensillary areas (Figs 4, 21–25, 27–31, 36, 38, 42, 80–82, 85–91, 93–94, 97–100, 111–115, 120–121, 126–137, 139–147, 149–152, 154–157, 160–175, 177, 182, 186–189, 207–208, 216).

7) sensory cavities that are sensory organs totally or partially invaginated into the body of the antennomere, with only a narrow slit remaining on the outside (Figs 176, 178, 180–181, 184, 190, 197–200, 204) or being completely closed (Figs 179, 183, 185, 201–203).

Sensory organs of types 4–7 have approximately the same sensillary composition as internal B4a, B4b (sometimes only one of them), M5, rarely M6, and usually C7 types having a fixed apical or subapical position, more rarely occurring on the sides of antennomeres. Sensory fields (3rd type) usually occupy the sides and inner margins, sometimes also forming depressions in cuticle and transforming into lateral pits and fossae; in this way the latter differ from apical organs in having typical or modified B4c-type sensillae exclusively. The types and arrangements of sensory organs may serve as reliable taxonomical characters, but it is important to note that organs that appear superficially similar may actually belong to different morphoclines, i.e. originate in different ways that provide no evidence for phylogenetic relations.

II. List of examined buprestid higher taxa arranged with respect to antennal structures

This study is based on generic rank taxa. The following list is made up of only buprestid taxa studied using a scanning electron microscope. The arrangement and status of suprageneric taxa reflect similarities and presumed evolutionary trends in antennal structures rather than following any formal classification. Since there is no agreement regarding the composition and status of the higher taxa, I use such informal categories as complex, lineage, branch, and generic group, which frequently do not coincide with formal taxa. Descriptions of antennal structures of separate genera or generic groups are presented in the chapter "III. A review of the antennal structures in Buprestidae"; explanations of hypothetical morphoclines and evolutionary trends are provided in the chapter "IV. Evolution and general trends in transformation of antennal structures in Buprestidae"; systematic changes and proposals are discussed in the chapter "V. Taxonomical implications".

Schizopoid complex (Schizopodinae LE CONTE, 1861)

Schizopodini LE CONTE, 1861

Schizopus LE CONTE

Dystaxiini THÉRY, 1929

Glyptoscelimorpha (s.str.) HORN
G. (*Distaxiella* KNULL)
Dystaxia LE CONTE

Julodoid complex (Julodinae LACORDAIRE, 1857)

Julodini LACORDAIRE, 1857

Aaata SEM.
Sternocera ESCHSCH.
Julodis ESCHSCH.
Julodella SEM.
Neojulodis KERR. (*Protojulodis* HOLM)
Amblysterna SAUND.

Polycestoid complex (Polycestinae LACORDAIRE, 1857)

Acmaeoderioid lineage

Mastogeniini LE CONTE & HORN, 1883

Mastogenius SOL.
Micrasta KERR.

Acmaeoderini KERREMANS, 1893 (sensu novo)

Odetteina **subtr.nov.**

Odettea BAUD.

Nothomorphina COBOS, 1955

Nothomorphoides HOLM
Nothomorpha SAUND.

Acmaeoderoidina COBOS, 1955

Acmaeoderoides VAN DYKE

Acmaeoderina KERREMANS, 1893

Acmaeodera (s.str.) ESCHSCH.
A. (*Palaeotethya* VOLK.)
A. (*Acmaeotethya* VOLK.)
A. (*Cobosiella* VOLK.)
A. (*Paracmaeodera* THÉRY)
A. (*Ptychomus* MARS.)

A. (*Rugacmaeodera* HOLM)
Acmaeoderopsis BARR
Anambodera BARR
Microacmaeodera (s.str.) COBOS
Xantheremia (s.str.) VOLK.
Acmaeoderella (s.str.) COBOS
A. (*Carininota* VOLK.)
A. (*Euacmaeoderella* VOLK.)

Ptosimini KERREMANS, 1902

Ptosima DEJ.

Paratrachyini COBOS, 1980

Sponsor (s.str.) LAP. & GORY
Paratrachys SAUND.

Polyctesioid lineage

Thrincopygini LE CONTE, 1861

Thrincopyge LE CONTE

Chrysophana generic group

Chrysophana LE CONTE
Beerellus NELS.

Polyctesini COBOS, 1955

Schoutendeiastes BOURG.
Bellamyina BÍLÝ
Paraxenopsis COBOS
Polyctesis MARS.

Polycestioid lineage

Bulis generic group (Bulina BELLAMY, 1995)

Bulis LAP. & GORY

Polycestini LACORDAIRE, 1857

Strigopteroides COBOS
Strigoptera DEJ.
Jelinekia COBOS
Polycesta DEJ.
Paracastalia KERR.
Polycestina (s.str.) COBOS
P. (*Damarana* COBOS)
Pseudocastalia KRAATZ
Thurntaxisia Schatzm.

Tyndarini COBOS, 1955

Tylauchenina COBOS, 1973

Tylauchenia BURM.

Tyndarina COBOS, 1955)

Tyndaris (s.str.) THOMS.
T. (*Paratyndaris* FISH.)

Acherusina COBOS, 1955

Acherusia (s.str.) LAP. & GORY

Pseudacherusina COBOS, 1980

Pseudacherusia KERR.

Xyroscelini COBOS, 1955

Xyroscelis THOMS.

Prospherini Cobos, 1980
Prospheres Thoms.
Euleptodema Obenb.
Astraeusini Cobos, 1980
Astraeus Lap. & Gory

4. Chalcophoroid complex (Chalcophorinae Lacordaire, 1857)

Chalcophorioid lineage
Vadonaxiini Descarpentries, 1969
Vadonaxia Desc.
Paratassini Bílý & Volkovitsh, 1996
Paratassa Mars.
Nanularia generic group
Nanularia Casey
Ampheremus Fall
Chalcoplia generic group
Chalcoplia Saund.
Descarpentriesiola Cobos
Embrikillium Obenb.
Chalcophorini Lacordaire, 1857 (sensu novo)
Chrysochroina Laporte, 1835
Chrysochroa generic group
Asamia Théry
Chrysochroa (*Catoxantha* Dej.) Lap. & Carcel in Dej.
Agelia Lap.& Gory
Callopistus generic group
Epidelus Deyr.
Cyalithus Thoms.
Steraspis Dej.
Pygichaeta Obenb.
Chrysaspina Théry
Callopistus Deyr.
Micropistus Théry
Philocteanus Deyr.
Chalcophorina Lacordaire, 1857
Cyphogastra generic group
Cyphogastra Deyr.
Cyphogastrella Théry
Papuodema Obenb.
Paracupta Deyr.
Callistroma Fairm.
Sapaia generic group
Sapaia Bílý
Genus 1
Rhabdolona Obenb.
Scaptelytra generic group
Scaptelytra Saund.
Iridotaenia generic group
Iridotaenia Deyr. (*Iridomroczkowskia* Hoł.)
Parataenia Kerr.

Chrysodema generic group

Metataenia Théry (*Metamroczkowskia* Hoł.)
Chrysodema Lap. & Gory
Thymedes Waterh.
Tamamushia Miwa & Chujô
Nipponobuprestis Obenb.
Gelaeus Waterh.

Chalcophoropsis generic group

Chalcophoropsis Saund.

Texania generic group

Texania Casey

Chalcophorella generic group

Chalcophorella Kerr.
Stigmatophorella Obenb.

Pseudotaenia generic group

Pseudotaenia Kerr.

Chalcophora generic group

Chalcophora Dej.

Chlorophorella generic group

Chlorophorella Desc.

Evidini Toyama, 1987

Evides Serv. in Dej.

Hypoprasini Hołynski, 1993, **stat.nov.**

Euchromatina Hołynski, 1993

Euchroma Sol.

Hypoprasina Hołynski, 1993

Hypoprasis Fairm. & Germ.
Baudonisia Cobos
Cordillerita Obenb.

Pristipterina Hołynski, 1993

Halecia Lap. & Gory
Eupodalecia Obenb.

Pelecopselaphus generic group

Pelecopselaphus Sol.
Chrysesthes Sol.

Euplectaleciina Hołynski, 1993

Euplectalecia Obenb.

Cinyrina Cobos, 1979, **stat.nov.**

Cinyra Lap. & Gory

Saundersina generic group

Saundersina Cobos

Poecilonotini Alexeev & Bebka, 1970

Poecilonotina Alexeev & Bebka, 1970

Palmar Schaefer (s.str.)
P. (*Scintillatrix* Obenb.)
Poecilonota Eschsch.

Nesotrinchus generic group

Nesotrinchus Obenb.

Psilopterioid lineage

Sphenopterini Lacordaire, 1857

Sphenoptera Dej. (s.str.)
S. (*Chrysoblemma* Jak.)
S. (*Sphenopterella* Volk. & Kalash.)
Armenosoma Waterh.
Evagora Kerr.
Genestia Théry

Psilopterini Lacordaire, 1857

Hippomelanina Hołynski, 1993

Hippomelas Lap. & Gory (s.str.)

Pseudoperotina Toyama, 1987

Pseudoperotis Obenb.
Asidoptera Obenb.

Psilopterina Lacordaire, 1857

Psiloptera generic group

Achardella Obenb.
Pseudolampetis Obenb.
Ectinogonia Spin.
Chalcopoecila Saund.

Polybothris generic group

Polybothris Dup.

Lampetis generic group

Capnodis Eschsch.
Cyphosoma Mnnh.
Perotis Dej.
Lampetis Spin.
Oedisterna Lac.

Dicerca generic group

Latipalpis Sol. (s.str.)
L. (*Palpilatis* Bílý)
Dicerca Eschsch. (s.str.)
D. (*Argante* Gistel)

Dicercomorpha generic group

Touzalinia Théry
Dicercomorpha Deyr.

Haplotrinchina Hołynski, 1993

Cardiaspis Saund.
Haplotrinchus Kerr.

5. Buprestoid complex (Buprestinae Leach, 1815)

Mendizabalini Cobos, 1968

Mendizabalia Cobos

Philandia generic group

Philandia Kerr.

Maoraxiini Hołynski, 1984

Maoraxia Obenb.

Buprestioid lineage

Julodimorphini Kerremans, 1902

Julodimorpha Gemm. & Har.

Buprestinioid branch

Nascio generic group (Nascionina HOŁYNSKI, 1988)

Nascioides KERR.
Nascio LAP. & GORY

Pygicera generic group

Pygicera KERR.

Epistomentini LEVEY, 1978

Epistomentis SOL.
Diadoxus THOMS.
Cyria SOL.
Xenocyria OBENB.

Neobuprestis generic group

Neobuprestis KERR.
Balthasarella OBENB.

Buprestini LEACH, 1815

Trachykelina HOŁYNSKI, 1988

Trachykele MARS.

Buprestina LEACH, 1815

Buprestis L. (s.str.)
B. (*Orthocheira* RICHTER)
Eurythyrea DEJ.
Yamina KERR.

Lamprocheilina HOŁYNSKI, 1993

Lamprocheila SAUND.

Agaeocerina NELSON, 1981

Mixochlorus WATERH.
Peronaemis WATERH.
Agaeocera SAUND.

Exagistini TOYAMA, 1987

Exagistus DEYR.

Phrixiini COBOS, 1975

Paraphrixia SAUND.
Phrixia DEYR.

Stigmoderinioid branch

Buprestina generic group

Buprestina OBENB.

Bubastini OBENBERGER, 1920

Euryspilus LAC.
Bubastes LAP. & GORY

Microcastalia generic group

Microcastalia HELL.

Stigmoderini LACORDAIRE, 1857

Dactylozodes CHEVR.
Hiperantha GISTEL (s.str.)
H. (*Hyperantella* HOSCH.)
Conognatha ESCHSCH.
Calodema GORY & LAP.
Themognatha SOL.
Stigmodera ESCHSCH.
Hypostigmodera BLACKB.

Aglaostola branch

Aglaostola generic group

Aglaostola Saund.

Hilarotes generic group

Hilarotes Saund.

Spectralia generic group

Spectralia Casey
Ditriaena Waterh.

Pterobothrini **trib.nov.**

Pterobothris Fairm. & Germ.

Anthaxioid lineage

Thomassetiinioid branch

Neobubastes generic group

Neobubastes Blackb.
Eububastes Obenb.

Thomassetiini Bellamy, d’Hotman & Holm, 1987

Thomassetia Théry
Augrabies Bell.
Jakovleviola Obenb.

Aristosoma generic group

Aristosoma Saund.

Philanthaxia generic group

Philanthaxiina Hołynski, 1988

Philanthaxia Deyr.

Kurosawaia generic group

Kurosawaia Toyama & Ohmomo

Kisanthobiini Richter, 1949

Kisanthobia Mars.

Anthaxiinioid branch

Trigonogenini Cobos, 1956

Trigonogenium Har.

Coomaniellini Bílý, 1974

Coomaniella Bourg.

Curis generic group (Curidina Hołynski, 1988)

Curis Lap. & Gory
Neocuropsis Obenb.

Neocuris generic group (Neocuridina Hołynski, 1988)

Neocuris Saund.

Torresita generic group

Torresita Gemm. & Har.
Pseudanilara Théry
[*Neotorresita* Obenb.]

Theryaxia generic group

Theryaxia Cart.

Anilara generic group

Anilara Saund
Anthaxoschema Obenb.
Notographus Thoms.

Melobasis generic group

Melobasis LAP. & GORY (s.str.)
M. (*Briseis* SAUND.)
Montrouzieretta OBENB.

Anthaxiini LAPORTE & GORY, 1837

Cylindrophora generic group

Cylindrophora SOL.
Paracuris OBENB.
Tetragonoschema THOMS.

Anthaxiina LAPORTE & GORY, 1837

Chalcogenia SAUND.
Anthaxia ESCHSCH. (s.str.)
A. (*Haplanthaxia* REITT.)
A. (*Melanthaxia* RICHTER)
Cratomerus SOL. (s.str.)
C. (*Cryptocratomerus* RICHTER)
Brachelytrium OBENB.
Brachanthaxia THÉRY

Xenorhipini COBOS, 1986

Xenorhipis generic group

Xenorhipis LE CONTE
Hesperorhipis FALL

Trichinorhipis generic group

Trichinorhipis BARR

Chrysobothrioid lineage

Melanophilinioid branch

Melanophilini BEDEL, 1921

Juniperella generic group

Juniperella KNULL

Melanophila generic group

Xenomelanophila SLOOP
Phaenops DEJ.
Melanophila ESCHSCH. (s.str.)
Trachypteris KIRBY

Merimna generic group

Merimna SAUND.

Chrysobothrinioid branch

Cromophila generic group

Cromophila COBOS

Actenodini KERREMANS, 1893

Pseudactenodes KERR.
Belionota ESCHSCH. (s.str.)
Actenodes DEJ.

Chrysobothrini LAPORTE & GORY, 1837

Chrysobothris ESCHSCH. (s.str.)
C. (*Abothris* SEM. & RICHTER)
Colobogaster SOL.
Afrobothris THÉRY

Incertae sedis

Fahraeusia OBENB.

6. Galbelloid complex (Galbellinae REITTER, 1911)

Galbellini REITTER, 1911
Galbella WESTW.

III. A review of the antennal structures in Buprestidae

Schizopoid complex

Schizopus (Fig. 1) (see also NELSON & BELLAMY 1991: Figs 14–15, 22–23). Antennomeres: 11; 5–10th transverse triangular, thickened, 11th irregularly spherical or slightly elongated. Dimorphism: antennomeres enlarged in males. Apical organs: poorly defined depressions on anterior surface of 5–10th (Fig. 1, AD) and on both sides of 11th, lateral organs lacking. Dense, short A3 and coeloconic B4c sensillae cover the surface uniformly; sparse U1 sensillae arranged singly almost at the same level near antennomere apices and in small groups on apical corners.

Dystaxia, *Glyptoscelimorpha*. (Figs 2–4) (see also NELSON & BELLAMY 1991: Figs 9–13, 16–21). Antennomeres: 12; 5–11th poorly to strongly elongated, thickened (*Glyptoscelimorpha*, Fig. 2) or elongate triangular and strongly bilaterally flattened (*Dystaxia*, Fig. 4); 12th elongated. Dimorphism: poorly developed in some species, whereas in others the male antennomeres are abruptly enlarged. Sensory organs: poorly defined subapical depressions (*Glyptoscelimorpha*, Fig. 2) or subapical and lateral separate pits and fossae (*Dystaxia*, Fig. 4) on both sides of 5–11th; internal M5 and C7 sensillae also situated outside sensory organs; 12th has no distinct organs or has barely marked depressions internally. Dense, short A3, and coeloconic B4c sensillae regularly cover most of the surface; sparse U1 sensillae arranged singly almost at the same level near apices of antennomere (Fig. 2) and in small groups on apical corners.

Julodoid complex

Julodini (Figs 5–9). Antennomeres: 11; 5–10th transverse to slightly elongate triangular, strongly bilaterally flattened; 11th vary in the shape, usually elongated. Dimorphism not developed. Sensory organs (Figs 6, SZ; 7): poorly defined zones containing internal B4a, B4b, M5, M6, and C7 sensillae located along the glabrous outer margins and near the anterior margins on internal side of 5–11th almost completely concealed by very dense external sensillae; *Aaata* also bears small sensillary pits on glabrous outer margin (Fig. 6, SP). Lateral organs: extensive fields of very dense, short, modified, often apically bifurcated A3 and styloconic B4c sensillae, completely covering both sides of 5–11th, leaving only outer margins glabrous; these fields sometimes consist of sparser and shorter sensillae along the inner (*Julodis*, *Neojulodis*, *Julodella*) or outer margins near the apices (*Aaata*, Fig. 8); *Sternocera* (Fig. 9) has fields of modified, unguiculate, smooth-walled sensillae (possibly S8 in the male) in combination with B4c types delimited from adjacent fields of A3 sensillae. Sparse U1 sensillae occur singly on the sides; short thick sensillae, possibly derived from U1 types, occur in small groups on apical corners; normal A3 sensillae concentrate on glabrous zones along the outer margins and at the apex of 11th (Fig. 5).

Polycestoid complex

Antennomeres: 11, except *Acmaeoderella* (*Kocheridia*) and some *Mastogenius* which have 10 antennomeres; their shapes and the structure of sensory formations vary in different groups. Normal or slightly shortened U1 sensillae occur singly or in small groups on apical corners, anterior part of 11th, sometimes incorporated into lateral fields; long, often squamoid A3 sensillae concentrate on glabrous areas.

Mastogenius, *Micrasta* (Figs 10–11). Antennomeres 4–10 conical to elongate triangular, thickened; 11th elongate ovoid. Dimorphism: male antennomeres (Fig. 11) distinctly lengthened, bearing long trichoid sensillae evenly dispersed; females' (Fig. 10) mainly glabrous, covered with long aporous sensillae. Apical organs: defined zones comprised of elongated styloconic internal B4a, B4b, and M5 sensillae on anterior surface and corners of 4–10th, and on both sides of 11th (Fig. 10); lateral organs lacking. Antennomere surface mainly glabrous; bearing few U1 and B4c sensillae adjacent to sensory zones, as well as long A3 sensillae (Fig. 10).

Odettea, *Ptosima*, *Nothomorphoides*, *Nothomorpha*, *Acmaeoderoides*, *Acmaeodera*, *Acmaeoderopsis*, *Anambodera*, *Microacmaeodera*, *Xantheremia*, *Acmaeoderella* (Figs 12–18). Antennomeres 5–10 transverse to elongate triangular, thickened or poorly bilaterally, sometimes transversely, flattened; 11th varies in shape, usually elongated. Dimorphism: occasionally male antennomeres strongly enlarged (Fig. 14) or transformed (Fig. 17) and covered with dense basiconic sensillae, whereas closely related species often exhibit no dimorphism at all. Extreme dimorphism is observed in *Acmaeoderella* (*Euacmaeoderella*) *subcyanea* (Figs 17–18) in which the male distal antennomeres are strongly transversely enlarged and shifted around transverse axes, almost forming a pectinate flagellum (Fig. 17); the males of *Microacmaeodera longicornis* (COBOS) have strongly elongated antennomeres resulting in the antennae reaching halfway along the body. Apical organs: zones or poorly developed depressions on anterior surface of 4(5)–11th, containing B4a, B4b, M5, and C7 sensillae; lateral organs absent. B4c sensillae dispersed on both sides of expanded inner part (Figs 12, 14, 16–18) or concentrated near apical corners, outer part or large areas of antennomeres glabrous (Figs 13, 15).

Sponsor, *Paratrachys* (Figs 19, 20). Antennomeres 5–10 transverse to elongate triangular, thickened or poorly bilaterally flattened; 11th elongated. Apical organs: defined, slightly elevated zones containing long styloconic B4a, B4b, M5, and C7 sensillae on anterior surfaces or corners of 5–10th and on top of 11th; lateral organs absent; few B4c sensillae occur outside the sensory zones, surface glabrous.

Thrincopyge (Figs 32–34). Antennomeres 5–10 serrate-truncate, strongly bilaterally flattened; 11th rhomboid (Fig. 32). Sensory organs: poorly defined subapical zones on internal surface, from 5th onwards; these zones containing B4a, B4b, M5 and C7 sensillae almost completely concealed by very dense, long A3 sensillae (Figs 32, 34). Lateral organs: extensive fields of B4c, dense A3, and sparse, short U1 sensillae (Fig. 33) on both sides; outer margins and the greater part of sides glabrous.

Chrysophana, Beerellus (Fig. 40). Antennomeres 4–10 serrate-truncate, bilaterally flattened; 11th elongated. Apical organs: well defined, almost regular, slight internal shift of fossae on anterior surfaces of 5–10th, and on both sides of 11th; lateral organs: extensive fields of coeloconic B4c sensillae along inner margins on both sides, remaining part mainly glabrous.

Schoutendeiastes, *Bellamyina*, *Paraxenopsis*, *Polyctesis* (Figs 35–39) differ significantly in antennomere shape and the level of sensory organ differentiation. *Schoutendeiastes* (Fig. 35): antennomeres 5–10 serrate-truncate or transverse triangular, poorly bilaterally flattened, 11th elongated; dimorphism: male antennomeres enlarged; apical organs: poorly defined, slight internal shift of depressions on anterior surfaces of 4–10th and on both sides of 11th; lateral organs: extensive fields of long B4c sensillae leaving the greater part of antennomeres glabrous. *Bellamyina* (Fig. 36): antennomeres 4–10 triangular, poorly bilaterally flattened, 11th relatively short; apical organs: well defined, nearly circular, slight internal shift of fossae on anterior surfaces of 4–10th and on both sides of 11th; lateral organs: fields of long B4c, short U1, and long A3 sensillae along the inner margins on both sides, leaving the greater part of antennomeres glabrous. *Paraxenopsis* (Fig. 37): antennomeres 5–10 subquadrate with straightly cut inner margins forming a complete line, strongly bilaterally flattened, 11th elongated with regularly rounded apex; apical organs: moderately defined subapical fossae on internal side of 4–10th and on both sides of 11th; lateral organs: fields of long B4c and short U1 sensillae along the inner margins on both sides. *Polyctesis* (Figs 38–39): antennomeres 4–10 serrate-truncate, strongly bilaterally flattened, 11th elongated; apical organs: well defined, nearly circular subapical fossae on internal side of 4–10th and on both sides of 11th; lateral organs: fields making up anterior part comprising U1 and long B4c sensillae, posterior with short B4c sensillae along the inner margins.

Bulis (Fig. 21). Antennomeres 4–10 elongate triangular, bilaterally flattened; 11th elongate ovoid. Sensory organs: large, round, subapical fossae on internal side of 4–10th and on both sides of 11th; lateral organs absent; B4c and long A3 sensillae nearly uniformly dispersed.

Jelinekia, *Paracastalia*, *Thurntaxisia*, *Pseudocastalia*, *Strigoptera*, *Strigopteroides*, *Polycesta*, *Polycestina*, (Figs 22–27). Antennomeres 4–10 vary in shape, 11th usually elongate ovoid. Dimorphism: not developed, antennomeres in males nearly as large as in females. Sensory organs: vary in shape, large, deep, often irregular subapical, more rarely both subapical and lateral, sometimes confluent fossae, located on anterior surface, internal or both sides beginning from 4–5th. Superficial B4c sensillae (if present) occur near the sensory organs. Surface mainly glabrous, sensillae on U1 and A3. *Strigopteroides* (Fig. 26): antennomeres 4–10 subcylindrical to elongate conical; sensory organs: poorly defined irregular subapical depressions and groups of external B4c sensillae begin from 4th, located on sloping anterior margins; 11th with depression on each side. *Strigoptera*: antennomeres a little more flattened; sensory organs: deep confluent depressions and fossae containing unified B4b sensillae. *Jelinekia* (Fig. 22): antennomeres 5–10 transverse triangular with very large irregular fossae internally, containing well defined, long B4a, B4b, B4c, M5, and C7 sensillae; 7–10th with small fossae externally. *Polycesta* (Fig. 27): antennomeres 5–10 elongate triangular, 4th nearly subcylindrical, bearing small, deep, more or less regular circular or ovoid, separate apical and lateral fossae, containing unified B4b sensillae. *Paracastalia*, *Polycestina*, *Pseudocastalia*, and *Thurntaxisia* (Figs 23–25): antennomeres bear apical or subapical, circular, deep, small fossae on both or only internal side containing unified B4b sensillae.

Tylauchenia, *Neocypetes*, *Tyndaris*, *Acherusia*, and *Pseudacherusia* (Figs 28–31). Antennal structures are very similar to those in previous group; apical organs well defined, appear from 5th or following (6–9th) antennomeres with slight internal shift, 11th with depression on each side, lateral organs absent; external B4c sensillae demonstrate a tendency to incorporate into apical organs (Fig. 31, B4c); surface mainly glabrous. *Tylauchenia* (Fig. 28), *Neocypetes*: antennomeres 3–5(6) subcylindrical, 6(7)–10 triangular; nearly regular apical fossae and groups of B4c sensillae appearing from 6–7th. *Tyndaris* (Fig. 29): antennomeres 5(8)–10 transverse to elongate triangular; circular apical fossae and groups of B4c sensillae, sometimes deepened into shallow depressions, appear from 5–8th. *Acherusia*: antennomeres 6–10 transverse triangular; transverse apical fossae and groups of B4c sensillae appearing from 6th. *Pseudacherusia* (Figs 30–31): antennomeres 6–10 transverse triangular; transverse apical fossae, which appear from 6th, containing not only unified B4b sensillae but also external B4c types (Fig. 31); surface glabrous, bearing only very sparse A3 sensillae.

Xyroscelis (Fig. 41). Antennomeres 4–10 elongate triangular, slightly flattened, 11th strongly elongated. Apical organs: poorly defined, round, internally shifted, subapical depressions comprising styloconic B4a, B4c, M5, and C7 sensillae on anterior corners of 4–10th and on both sides of 11th; lateral organs: groups of long B4c and shortened U1 sensillae adjacent to subapical organs. Surface mainly glabrous, bearing sparse A3 sensillae.

Prospheres (Fig. 43), *Euleptodema*. Antennomeres 4–10 serrate-truncate, strongly flattened, 11th irregular. Apical organs: poorly defined, large, round subapical depressions containing numerous styloconic basiconic sensillae located on internal side of 4–10th and on both sides of 11th; lateral organs: extensive fields of styloconic B4c sensillae occupying the greater part of the surface.

Astraeus (Fig. 42). Antennomeres 4–10 serrate-truncate, slightly widened towards apex, flattened, 11th ovoid. Dimorphism: males of subgenus *Astraeus* (s.str.) have strongly elongated antennomeres (Barker 1975). Apical organs: well defined, small, circular subapical fossae containing numerous styloconic sensillae, located at anterior corners on internal side of 4–10th and on both sides of 11th; lateral organs: elongated fields of styloconic B4c sensillae in shallow depressions on internal side.

Chalcophoroid complex

Antennae with 11 antennomeres, with rare exceptions (*Philocteanus* has 10 antennomeres as a result of fusing 10th and 11th). Sexual dimorphism occurs occasionally.

Vadonaxia (Fig. 44). Antennomeres 4–10 transverse triangular, strongly flattened, 11th irregular. Apical organs: irregular depressions and poorly defined fossae at anterior margins on internal side of 4–10th, sometimes confluent with lateral organs; 11th with poorly defined depressions on both sides; containing B4a, B4c, and M5 sensillae, frequently situated outside apical organs; lateral organs: poorly defined zones and depressions comprising the same sensillae but those of B4c type predominating. Outer part of antennomeres mainly glabrous. U1 sensillae occur singly or in small groups on apical corners and anterior part of 11th; long A3 sensillae concentrated along the outer margins.

Paratassa (Fig. 45) (see also Bílý & Volkovitsh 1996: Figs 46–47). Antennomeres 4–10 transverse triangular, poorly bilaterally, sometimes strongly transversely flattened; 11th irregular. Dimorphism: male antennomeres enlarged and transversely flattened (Fig. 45), entire antennae elongated. Apical organs: poorly defined irregular depressions resulting from the junction of smaller ones, located on anterior surface (male) or internal side (female); containing B4a, B4b, and M5 sensillae, which also occur inside adjacent depressions. Antennomere surface covered with deep pores or small pits showing a tendency to confluency towards apex and containing single or sparse B4c sensillae, leaving only outer margins glabrous; single U1 sensillae occur on apical corners and at the apex of 11th.

Nanularia, *Ampheremus* (Figs 46–47) (see also Bílý & Volkovitsh 1996, Figs 48–51). Antennomeres 4–10 rounded triangular (*Nanularia*, Fig. 47) or transverse (*Ampheremus*, Fig. 46), thickened; 11th irregular, appendiculate owing to transverse depression separating its apex. Dimorphism: males of *Nanularia* have enlarged antennomeres and different colours (Bellamy 1987). Apical organs: large, transverse, poorly (*Ampheremus*) or well (*Nanularia*) defined depressions on anterior surface comprising B4a, B4b, and M5 sensillae. Antennomere surface covered with deep pores containing single B4c sensillae leaving outer part glabrous, covered with sparse A3 sensillae; U1 sensillae occur singly around the apex.

Chalcoplia (Fig. 48). Antennomeres 4–10 rounded triangular, thickened; 11th irregular. Apical organs: small, deep, defined depressions located asymmetrically on anterior surface of 4–10th and on the both sides of 11th, containing B4a and M5 sensillae. Antennomere surface covered with deep pores having a tendency to confluency, leaving outer part glabrous apart from sparse A3 sensillae; U1 sensillae occur singly around the apex.

Descarpentriesiola (Figs 49, 50). Antennomeres 4–10 slightly transverse triangular, poorly flattened; 11th irregularly ovoid. Apical organs: poorly defined, confluent depressions containing B4a, B4c, M5, and C7 sensillae in the middle of anterior surfaces of 4–10th and near the apex of 11th; lateral organs: frequently confluent, irregular depressions and pores containing B4c sensillae on both sides, leaving outer parts glabrous. Shortened U1 sensillae occur in small groups on apical corners and singly at the apex of 11th; A3 sensillae dispersed over glabrous outer surface.

Embrikillium (Fig. 51). Antennomeres 4–10 serrate-truncate, flattened; 11th ovoid. Apical organs: confluent irregular fossae containing B4a, B4c, M5, and C7 sensillae on anterior surfaces of 7–10th, and subapical fossae on 4–6th and 11th; lateral organs: frequently confluent depressions, fossae and pores containing B4c sensillae on both sides, leaving outer parts glabrous. Numerous U1 sensillae located in groups at apical corners and along anterior margin of 11th; A3 sensillae dispersed over glabrous outer surface.

Asamia, *Chrysochroa*, *Agelia* (Figs 52–53). Antennomeres 4–10 strongly transverse triangular or pectinate (*Agelia*, Fig. 53), sometimes attached at an angle to longitudinal (some *Chrysochroa*) or transverse (*Agelia*) antennal axes; 11th ovoid or irregular. Dimorphism: male antennomeres sometimes slightly enlarged. Fixed sensory organs absent, sometimes apical or lateral ones occur as a result of the confluence of small pores containing primarily single or a few basiconic sensillae; pores more or less uniformly cover the surface from 4th onwards, leaving only outer margins glabrous. U1 sensillae of variable length occur singly or in small groups at the apical corners and anterior margins; sparse A3 sensillae, mainly concentrated along the outer margins.

Callopistus, *Micropistus* (Fig. 54), *Steraspis*, *Pygichaeta*, *Chrysaspina*, *Epidelus*, *Philocteanus*, *Cyalithus*. Antennomeres 3(4)–(9)10 slightly elongate to sharply transverse triangular, pores appear from 3rd onwards. *Epidelus* antennomeres serrated from 4th, although there is a poorly visible area of pores on the internal side of 3rd. Sensory organs are very similar to those in previous group. There is a tendency for antennomeres 10–11 to be partly or complete fused, most marked in *Micropistus* and *Philocteanus*. Another trend is towards the differentiation of fields occupying only the expanded parts of antennomeres, leaving the outer parts glabrous.

Cyphogastra, *Cyphogastrella* (Fig. 61), *Callistroma*, *Paracupta*, *Papuodema*. Antennomeres 4–10 strongly elongate triangular or serrate-truncate, poorly flattened; 11th ovoid. Sensory pores and pits deep, mainly circular, separated from each other, almost completely covering the surface and leaving only the very outer margins glabrous.

Sapaia, Genus 1, *Rhabdolona* (Figs 56–58). Antennomeres 4–6 elongated, 7–10 triangular or serrate-truncate, 11 ovoid or irregular. Sensory pores almost completely confluent on 4–6th forming large single depression (Fig. 58); those on 7–11th form irregular apical and lateral depressions. Sensillae of 4B type in apical depressions are well defined (*Sapaia*) or unified (other genera).

Scaptelytra (Fig. 59) shows a similar trend for sensory pores to become confluent in large depressions but differs in having distinctly elongated distal antennomeres.

Iridotaenia (Fig. 55), *Parataenia*. Antennomeres 4–10 elongate triangular or serrate-truncate, flattened; 11th elongate ovoid. Apical organs (if present): large subapical fossae on internal side of 4–10th, and on both sides of 11th; lateral organs: pores and circular pits of different sizes on both sides which cover the surface, leaving only outer margins glabrous. Long U1 sensillae situated singly or in small groups at the apical corners and anterior margins; sparse A3 sensillae concentrated mainly along the outer margins. This genus is apparently heterogenous since the above-mentioned antennal structure looks much the same as in the *Chrysodema* group whereas that of some other species resembles the *Callopistus* group.

Metataenia, *Chrysodema*, *Thymedes*, *Tamamushia* (Fig. 63), *Gelaeus*, and *Chalcophoropsis* (Fig. 64). Distal antennomeres as in previous group but more strongly elongated and flattened; sensory pores and pits occupying both sides, leaving inner and outer margins glabrous.

Nipponobuprestis (Fig. 62) and *Texania* bear transverse apical depressions of confluent pores on distal antennomeres, similar to *Chalcophorella* and *Stigmatophorella*.

Chalcophorella, *Stigmatophorella* (Fig. 60). Antennomeres 4–10 triangular, 11th irregularly ovoid. Sensory pores confluent into distinct, usually transverse apical depression and sometimes several lateral ones.

Chalcotaenia, *Austrophorella*, *Afrophorella* (studied with only optical microscopes), and *Pseudotaenia* have pits and pores concentrated into poorly defined, longitudinally extended lateral fields, but *Pseudotaenia* bears longitudinal zones of shortened U2 sensillae along inner margins, similar to *Chalcophora*.

Chalcophora (Figs 65–66). Antennomeres 4(5)–10 elongate serrate-truncate, strongly flattened; 11th elongate ovoid. Apical organs absent; lateral organs: longitudinally extended fields (Fig. 65, SF) of very deep, circular, widely separated pores and small pits comprising deepened unified B4b sensillae (Fig. 66) on both sides, leaving inner and outer margins glabrous. Shortened U1 sensillae produce well defined longitudinal zones between the fields and inner margins (Fig. 65, U1); A3 sensillae concentrated along outer margins.

Chlorophorella (Fig. 67). Antennomeres 4–10 sharply triangular, poorly bilaterally flattened; 11th irregular. Apical organs: small, deep, irregular depressions on anterior surface of 4–10th, and on both sides near the apex of 11th; containing a few B4a sensillae; lateral organs: fields of small pores containing single or a few B4c sensillae on both sides along the inner margins, leaving the greater part of the antennomere glabrous. Short U1 sensillae occur in groups on the apical corners and singly around anterior margins; very sparse A3 sensillae located mainly along the outer margins.

Evides (Figs 68–72). Antennomeres 4–10 triangular to strongly transverse triangular, thickened, 11th irregular; distal antennomeres asymmetrical, transformed.The shape and disposition of the sensory organs differ greatly in different species. Dimorphism remains to be researched. Sensory organs: extensive, distinctly delimited from the rest of surface, asymmetrical depressions containing strongly elongated, unified B4b and M5 sensillae; these appear on antennomere 4 as elongate lateral depressions (Fig. 72) and round apical fossae, then external depressions shift towards the inner margin and internal ones become confluent with apical fossae to form an extensive single depression. *E. gambiensis* (Figs 70–72) has no distinct apical fossae distally whereas *E. kraatzi* (Figs 68–69) has these on at least 8–10th. In *E. kraatzi* external depressions on antennomeres 7–11 asymmetrically invaginated (Fig. 69). The rest of antennomere surface glabrous, bearing sparse A3 sensillae; modified U1 sensillae occur in small groups on apical corners.

Euchroma (Fig. 73). Antennomeres 4–10 strongly transverse triangular with sharpened apical corners, thickened; 11th irregular. Distinct sensory organs absent, antennomere surface uniformly covered with deep, separated pores and small pits containing single or a few basiconic sensillae. Shortened U1 sensillae occur in groups on apical corners and singly along inner margins, on the sides, and on top of 11th; short, sparse A3 sensillae concentrated along outer margins.

Hypoprasis (Fig. 74), *Baudonisia*, *Cordillerita*. Antennomeres 3–10 elongated, nearly serrate-truncate with sharpened apical corners, thickened, sides poorly flattened; 11th irregularly ovoid. Sensory organs: longitudinal fields of confluent pores and irregular pits on both sides of 4–11th; larger subapical pits containing B4a, B4b, M5, and C7 sensillae; pores and lateral pits contain B4c types. U1 sensillae occur singly or in groups on apical corners and the apex of 11th; sparse A3 sensillae dispersed along glabrous outer margins.

Halecia, *Eupodalecia* (Figs 75–76). Antennomeres 4–6 triangular, 7–10 elongated, nearly serrate-truncate with round apical corners, poorly flattened; 11th irregularly ovoid. Sensory organs: pores and irregular pits concentrated into two big, often confluent subapical and lateral depressions on both sides of 4–11th; subapical depressions containing B4a, B4b, M5, and C7 sensillae; lateral ones containing only B4c sensillae. U1 sensillae occur singly along inner margins and on top of 11th; long A3 sensillae dispersed over glabrous areas.

Chrysesthes (Fig. 77), *Pelecopselaphus*. Antennomeres 3(4)–6 triangular, 7–10 transverse triangular with rounded apical corners, thickened; 11th irregular. Apical organs: triangular, widest at the base and narrowing towards the apex, well defined depressions on anterior surfaces of 4–10th, containing B4a, B4b, M5, and C7 sensillae. Lateral organs: extensive, poorly defined depressions of B4c sensillae on both sides; the septae between apical and lateral organs bear pores containing single B4c sensillae. U1 sensillae occur in groups on apical corners and on top of 11th; sparse A3 sensillae concentrated along glabrous outer margins.

Euplectalecia (Figs 78–79) has generally the same antennal structure as that found in *Halecia*-group, differing in the possession of distinct subapical fossae instead of depressions. Lateral depressions poorly defined, surrounded with pores; 11th bearing subapical and lateral depressions. External sensillae as in previous group.

Cinyra (Figs 80–81). Apical and lateral organs are well defined, round or ovoid fossae comprising unified basiconic sensillae; their locations on both sides are in complete agreement with that in *Euplectalecia*; remaining surface glabrous. Apex of 11th separated with transverse subapical fossae.

Saundersina (Fig. 82). Antennomeres 4–8 triangular, 9–10 transverse triangular with rounded apical corners, poorly flattened; 11th ovoid. Sensory organs: nearly circular or transverse-elliptical, well defined subapical fossae on internal side of 4–11th, containing unified B4b sensillae as well as M5 and C7 types. U1 sensillae occur singly along inner margins and on top of 11th; long, sparse A3 sensillae dispersed over glabrous surface.

Palmar, *Poecilonota* (Figs 83–87). Antennomeres 4–10 round, rarely sharp, elongate to transverse triangular, thickened or poorly flattened; 11th irregular. Apical organs: extensive zones on both sides (Figs 83–84), or very large, rounded fossae, internally shifted, on anterior surface (Figs 85–87); 11th bearing large fossa apically; lateral organs absent. Surface glabrous, bearing sparse A3 sensillae; long, sparse U1 sensillae occur singly on apical corners. *Palmar* (*Scintillatrix*) *limbata* (Figs 83–84) has extensive apical zones internally that are distinctly delimited from the rest of surface.These extend to external surface and consist of a poorly defined central part containing B4a, B4b, M5, and C7 sensillae surrounded with numerous coeloconic B4c types; 11th bearing small fossae and area of coeloconic sensillae anteriorly. *S. chinganensis* exhibits sensory zones more (male) or less (female, Fig. 85) defined, containing incorporated B4c sensillae; these do not extend to external surface. In *P.* (s.str.) *virgata* (Fig. 86) apical organs well defined, weakly transverse. Apical fossae of *Poecilonota* (Fig. 87) round, well defined, deeper, with denser U1 sensillae. *Nesotrinchus* (Fig. 88) exhibits nearly the same state of apical organs as *S. chinganensis* and *Poecilonota*.

Sphenoptera, *Armenosoma*, *Evagora*, *Genestia* (Figs 89–94; see also BÍLÝ & VOLKOVITSH 1996, Fig. 53). Antennomeres 4(5)–10 transverse to elongate triangular or serrate-truncate, moderately flattened; 11th varies widely in shape. Dimorphism: male antennomeres frequently enlarged. Apical organs: round, rarely irregular, deep, fossae usually internally shifted (Fig. 94, AF) containing B4a, B4b, M5, and C7 sensillae on anterior surface of 4–10th; 11th bearing subapical fossa internally. Lateral organs: fields of separated pores and small pits (Fig. 94, LF) of single or a few B4c sensillae on both sides, leaving outer parts glabrous. Sparse U2 sensillae (Fig. 94, U2) occur along inner margins forming no definite areas; U1 sensillae located on apical corners and on top of 11th; sparse A3 sensillae uniformly covering glabrous areas or concentrated along outer margins.

Hippomelas (Fig. 95). Antennomeres 4–10 elongate triangular to serrate-truncate, flattened; 11th elongated, appendiculate owing to transverse subapical fossae separating the very apex. Dimorphism: male antennomeres serrate-truncate, females' triangular. Apical organs: moderately defined, transverse depressions, internally shifted, containing B4a, M5, and C7 sensillae on anterior surface of 4–10th (Fig. 95, AD); 11th bearing subapical fossae internally; lateral organs: extensive fields of separated pores (Fig. 95, LF) of single B4c sensillae on both sides leaving outer parts glabrous. Dense U2 sensillae form definite longitudinal zones along inner margins (Fig. 95, U2), short U1 sensillae located on apical corners, A3 sensillae dispersed over glabrous areas.

Psilopterini (Figs 96–115). Antennomeres vary in shape from subcylindrical or conical (3rd–4th) and triangular (5–6th) to serrate-truncate (7–10th), flattened; 11th rhomboid, ovoid or irregular. Dimorphism: male distal antennomeres enlarged. Apical organs: vary significantly in different groups from irregular depressions internally shifted (Figs 101, 102) to regular deep fossae (Figs 97–99, 105, 108, 111–115) containing defined or unified B4a and B4b, as well as M5, and C7 sensillae on anterior surface of 6–10th; proximal (4–5th) and apical (11th) antennomeres bear single subapical fossa or several pits internally (Figs 97–99, 104, 107, 110). Lateral organs: extensive fields of separate or confluent pores, depressions and small pits (Figs 95, 96, 101–103, 105, 106), or several (Figs 97, 109) single (Figs 98–100, 104, 107, 108) large fossae containing basiconic sensillae on both sides or only on internal surfaces or even, rarely, absent. Dense U2 sensillae form definite longitudinal zones along inner margins, short U1 sensillae often occur singly in cuticular depressions at apical corners and the apex of 11th; A3 sensillae dispersed over glabrous areas.

Achardella (Fig. 96). Apical organs: well defined, deep fossae; lateral organs: poorly defined small pits and pores cover both sides uniformly, resembling *Hippomelas* (Fig. 95) and *Polybothris sulcicollis* (Fig. 103).

Asidoptera, *Pseudoperotis* (Figs 101–102). Apical and lateral organs: confluent, irregular depressions, fossae and pores with well defined internal basiconic sensillae;

external zones containing U2 and long B4c sensillae. *Polybothris sulcicollis* (Figs 103). Apical organs: transverse fossae; lateral fields consist of regular, confluent or separate pores, pits, and fossae; antennal structure resembling that of *Hippomelas* (Fig. 95).

Capnodis, *Cyphosoma*, *Latipalpis* (Figs 105–106, 109–110), and *Touzalinia*. Apical organs: regular, deep, round or transverse ovoid fossae; lateral fields well defined, consisting of more or less regular, separate pits and fossae.

Pseudolampetis, *Ectinogonia*, *Polybothris staudingeri*, *Perotis*, *Lampetis* (Figs 97–98, 104, 107–108), *Psiloptera* (Nelson 1982, Fig. 19), and *Oedisterna*. Apical organs: the same as in previous group; lateral organs: a few (Fig. 97) or single large depressions (Fig. 107) or fossae (Figs 98, 104, 108) on both sides of the antennomere.

Chalcopoecila (Figs 99–100). Antennomeres 4–10 transverse triangular with sloped anterior margins, 11th irregular. Apical organs: round, deep, internally shifted fossae containing basiconic and campaniform sensillae, located on anterior surface of 4–11th; lateral organs: the same as apical ones, located on both sides; the greater part of the surface glabrous; U2 sensillae arranged in small groups on apical corners, A3 sensillae concentrated along outer margins.

Dicerca (Figs 111–112). Antennomeres 4–10 rounded triangular, 11th irregular. Sensory organs: circular, deep subapical fossae containing unified B4b, as well as M5 and C7 sensillae (Fig. 112), located on internal side of 4–11th; lateral organs absent, the greater part of antennomeres glabrous; short U1 and U2 sensillae arranged along inner margins and on top of 11th; A3 sensillae occur along outer margins.

Dicercomorpha (Fig. 113). Antennal structure is very similar to *Dicerca*, but fossae larger and located on anterior surface, in many respects resembling Poecilonotini (Figs 85–87) and *Nesotrinchus* (Fig. 88) from which it differs in having distinct fields of U2 sensillae along inner margins. *Cardiaspis* and *Haplotrinchus* (Figs 114–115) have nearly the same antennal structure.

Buprestoid complex

Mendizabalia (Figs 116–117). Antennomeres 4–11 flabellate (male) or serrate (female, not studied), male antennomeres 4–10 transversely flattened. Defined sensory organs absent, whole surface uniformly covered with dense basiconic, possibly specialised male, and sparse aporous sensillae (Fig. 117, ?S8, A3); the sensory areas also bear internal sensillae on inner surface (Fig. 117, B4c, M5, C7); sparse, short U1 sensillae dispersed over the surface (Fig. 117, U1).

Philandia (Fig. 118). Antennomeres 4–10 serrate, rounded triangular, strongly bilaterally flattened, asymmetrical; 11th irregular. Dimorphism: distal antennomeres of male enlarged and bearing denser basiconic sensillae than those in female. Sensory organs: very large, occupying most of the antennomere surface, poorly defined, shallow subapical depressions (Fig. 118, SD), containing numerous B4a, B4b, and sparse M5 and C7 sensillae, located on internal face of 4–10th; lateral organs absent. Antennomere surface uniformly covered with B4c and A3 sensillae; short U1 sensillae located along the apices.

Maoraxia (Figs 213–215). Antennomeres 4–10 elongate (male) or rounded triangular (female), thickened; 11th elongate ovoid. Apical organs: zones or poorly defined, hardly visible, shallow depressions containing styloconic internal sensillae arising from well developed tubercles (Fig. 215, B4a, B4b, M5, C7), located on anterior surface of 4–10th and on both sides of 11th; lateral organs absent, inner margins bearing very long, dense B4c and shortened U1 sensillae that do not form distinct fields (Fig. 213). Antennomere surface mainly glabrous, covered with dense aporous sensillae (Figs 213–215, A3); U1 sensillae occur on inner margins as well as on anterior corners and on top of 11th. Antennal structure of *Maoraxia* is quite similar to that in the Mastogeniini (Figs 10, 11) in having internal sensillae arising from distinct tubercles and long, dense external ones.

Julodimorpha (Fig. 119). Antennomeres 4–10 elongate triangular with acute corners, bilaterally flattened; 11th narrowed, strongly elongated. Apical organs: poorly defined, irregular subapical depressions consisting of confluent fragments and containing B4a, M5, and C7 sensillae, located at anterior margins of 4–10th and on both sides of 11th; lateral organs: poorly defined depressions consisting of confluent smaller ones and containing short B4c sensillae on both sides. Antennomere surface glabrous with scattered A3 sensillae; short U1 sensillae occur at anterior margins and on top of 11th.

Nascio (Figs 161–162). Antennomeres 4–10 rounded triangular, slightly bilaterally flattened; 11th irregular. Apical organs: large, deep, internally shifted, nearly circular subapical fossae with narrow depressions adjacent to their inner margins (Fig. 162); 11th bearing circular fossa internally; containing B4a, M5, and C7 sensillae; lateral organs absent. Antennomere surface mainly glabrous, bearing very long basiconic (?B4c) sensillae which do not form definite zones, and long scattered A3 sensillae; long U1 sensillae occur at anterior corners and on top of 11th.

Nascioides (Fig. 160). Antennomeres 4–10 elongate triangular, slightly bilaterally flattened; 11th strongly elongated. Apical organs: large, slightly transverse, irregular fossae on anterior surface; 11th bearing circular fossa internally, containing internal B4a, M5, C7, and external B4c sensillae partly incorporated inside the fossae; lateral organs absent. Antennomere surface mainly glabrous, bearing long B4c and shortened U1 sensillae forming poorly defined zones near apical corners; long, normal and serrated A3 sensillae scattered along outer margins.

Pygicera (Fig. 120). Antennomeres 4–11 elongate triangular to serrate–truncate, poorly bilaterally flattened; 11th elongate ovoid. Apical organs: well defined, nearly circular, fossae internally shifted containing B4a, M5, and C7 sensillae, located at anterior margins of 4–10th and on both sides of 11th; lateral organs absent. Antennomere surface bears long B4c and A3 sensillae which do not form defined fields; long U1 sensillae occur at anterior and along inner margins and on top of 11th.

Diadoxus, *Epistomentis*, *Cyria*, *Xenocyria* (Figs 121–123). Antennomeres 4–10 elongate triangular to serrate-truncate, bilaterally flattened; 11th elongate ovoid or irregular. Apical organs: moderately defined, irregular, rarely regular, slightly or completely shifted to internal side, fossae at anterior margins of 4–10th and on both sides of 11th; containing B4a, M5, and C7 sensillae; lateral organs: extensive fields of short (*Diadoxus*: Fig. 121) or long (*Epistomentis*, *Cyria*: Fig. 122) B4c sensillae. Long U1 sensillae occur at anterior margins and on top of 11th; A3 sensillae concentrated mainly along glabrous outer margins. *Xenocyria* has strongly flattened antennomeres, small apical organs, and lateral fields partly deepened into indefinite depressions resembling those in Chalcophoroid taxa (Fig. 123).

Neobuprestis, *Balthasarella* (Figs 124–126). Antennomeres 3–10 (*Neobuprestis*) or 4–10 (*Balthasarella*) elongate triangular, bilaterally flattened; 11th elongated, irregular. Dimorphism (*Neobuprestis*): male antennomeres enlarged and almost completely covered with long basiconic sensillae (Fig. 124); those in female bear only fields of B4c sensillae along inner margins (Fig. 125); male of *Balthasarella* is unknown. Apical organs: poorly defined, irregular depressions or fossae internally shifted on anterior margins of 3(4)–10th and on both sides of 11th; containing B4a, M5, and C7 sensillae; lateral organs (females): fields of long B4c sensillae (Figs 125–126). Shortened U1 sensillae occur at anterior margins and on top of 11th; A3 sensillae dispersed over glabrous outer margins.

Trachykele (Fig. 127). Antennomeres 4–10 triangular, bilaterally flattened; 11th elongate ovoid, truncated apically. Apical organs: moderately defined, large, nearly circular fossae internally shifted at anterior margins of 4–10th and on both sides of 11th; containing B4a, M5, and C7 sensillae; lateral organs: extensive fields of B4c sensillae. Long U1 sensillae occur at anterior margins and on top of 11th; long, dense A3 sensillae dispersed over glabrous surface.

Buprestis, *Eurythyrea*, *Yamina* (Figs 128–130). Antennomeres 4–10 transverse to elongate triangular, bilaterally flattened, 11th ovoid or irregular. Apical organs: moderately defined, large, internally shifted, irregular fossae at anterior margins of 4–10th and on both sides of 11th, containing B4a, M5, and C7 sensillae; lateral organs: fields of short coeloconic (*Yamina*) or superficial (*Buprestis*, *Eurythyrea*) B4c sensillae (Figs 128, 130), those in *Eurythyrea* form indistinct, shallow depressions (Fig. 129). Long or slightly shortened U1 sensillae occur at anterior margins and on top of 11th; sparse A3 sensillae dispersed over glabrous surface.

Lamprocheila (Fig. 131). Antennomeres 4–10 triangular, bilaterally flattened; 11th elongate ovoid, truncated apically. Apical organs: defined, large, internally shifted, irregular fossae at anterior margins of 4–10th and on both sides of 11th, containing B4a, M5, and C7 sensillae; lateral organs: extensive fields of short and long styloconic B4c sensillae arising from tubercles. Long U1 sensillae occur at anterior margins and on top of 11th; A3 sensillae concentrated along glabrous outer margins.

Mixochlorus, *Peronaemis*, *Agaeocera* (Figs 135–138). Antennomeres 3–10 (*Agaeocera*: Figs 137, 138) or 4–10 (*Mixochlorus*, *Peronaemis*: Figs 135, 136) transverse to elongate triangular, with rounded anterior corners, bilaterally flattened; 11th irregular. Apical organs: moderately defined, large, internally shifted, irregular (*Mixochlorus*, *Peronaemis*) or regular (*Agaeocera*) fossae at anterior margins of (3)4–11th; containing B4a, M5, and C7 sensillae. Lateral organs: poorly defined, irregular depressions (*Mixochlorus*, *Peronaemis*) or well defined (*Agaeocera*) fossae on internal faces, containing B4c sensillae. Slightly shortened U1 sensillae occur at anterior margins and on top of 11th; sparse A3 sensillae dispersed over glabrous surface or concentrated along outer margins. Apical and lateral organs in *Peronaemis* demonstrate a tendency to become confluent into solitary, very large fossae divided by thin septa (Fig. 136). *Agaeocera* bears small additional pits and pores internally and along the inner margins (Figs 137–138).

Exagistus (Fig. 134). Antennomeres 4–10 serrate-truncate, bilaterally flattened. Apical organs: moderately defined, large, internally shifted, irregular fossae at anterior margins of 4–10th, containing B4a, M5, and C7 sensillae; lateral organs: extensive fields of short and long styloconic B4c sensillae arising from tubercles, forming large, indistinct lateral depressions separated from apical fossae by thin septa. Long U1 sensillae occur at anterior margins and on top of 11th; sparse A3 sensillae dispersed over glabrous surface.

Phrixia, *Paraphrixia* (Figs 132–133). Antennomeres 4–10 elongate serrate-truncate, bilaterally flattened. Apical organs: poorly defined, small, slightly shifted internally, nearly regular fossae at anterior margins of 4–10th; containing B4a, M5, and C7 sensillae; lateral organs: extensive fields of short and long B4c, and dense shortened U1 sensillae deepened into indistinct lateral depressions. U1 sensillae also occur at anterior margins and on top of 11th; sparse A3 sensillae dispersed over glabrous surface.

Buprestina (Fig. 139). Antennomeres 4–10 slightly transverse triangular, slightly bilaterally flattened; 11th irregularly rhomboid. Apical organs: large, deep, slightly transverse, internally shifted, irregular subapical fossae at anterior margins; these fossae are deeper at the base becoming shallower towards apical corners; 11th bearing circular fossae on both sides; containing unified B4a sensillae with long apical filaments and sparse M5 sensillae; lateral organs absent. Antennomere surface mainly glabrous, bearing B4c, long, scattered A3, and short ?U2 sensillae along inner margins forming no definite zones.

Bubastes, *Euryspilus* (Figs 140–142). Antennomeres 4–10 (*Bubastes*) or 6–10 (*Euryspilus*) slightly transverse to elongate triangular, with weakly sloping anterior margins, barely bilaterally flattened; 11th irregular. Apical organs: big, deep, slightly transverse, internally shifted, irregular subapical fossae at sloping anterior margins; these fossae are deeper at the base, becoming shallower towards apical corners; 11th bearing circular fossa apically; containing normal B4a or B4b, and sparse M5 sensillae; lateral organs absent (*Euryspilus*, Fig. 140) or represented by lateral fields (*Bubastes inconsistans*, Fig. 141) and poorly defined depressions (*B. iridescens*, Fig. 142) containing B4c sensillae. Antennomere surface mainly glabrous, bearing long, sparse A3 and short ?U2 sensillae.

Microcastalia (Fig. 143). Antennomeres 4–10 transverse triangular, with sloping anterior margins, barely bilaterally flattened; 11th irregular. Apical organs: big, deep, slightly transverse, irregular fossae on anterior surface; these fossae are deeper at the base, becoming shallower towards apical corners; 11th bearing circular fossae on both sides; containing B4a, M5, and C7 sensillae; lateral organs: small zones of short U2 and sparse B4c sensillae at apical corners. Antennomere surface glabrous, bearing sparse scattered A3 sensillae.

Dactylozodes, *Hiperantha*, *Conognatha*, *Calodema*, *Themognatha*, *Stigmodera* (Figs 144–149; see also GARDNER 1989, Figs 36–41). Antennomeres 3(7)–10 transverse or elongate triangular to serrate- truncate, bilaterally flattened; 11th variable in shape, usually irregular. Apical organs: large, deep, more or less regular fossae located on anterior surface, on sloping anterior margins, or on internal face; 11th bearing circular fossae apically, containing B4a, B4b, M5, and C7 sensillae; lateral organs: zones or fields of short U2 and B4c sensillae on apical corners, sometimes deepened into shallow depressions. Antennomere surface mainly glabrous, bearing scattered A3 sensillae. *Hypostigmodera* (Figs 148–149) differs greatly from other Stigmoderini in having 12 antennomeres and marked sexual dimorphism: male (Fig. 148) has flabellate antennae with antennomeres 4–11 bilobed, entirely covered with modified basiconic sensillae beginning from 4th, poorly defined small sensory depressions located at the base of distal antennomeres and at the apex of 12th; female (Fig. 149) has transverse triangular antennomeres, large sensory fossae located on anterior surface of 4–11th and near the apex of 12th, and the fields of short U2 and B4c sensillae along inner margins.

Aglaostola (Figs 150–151). Antennomeres 4–10 elongate triangular, poorly bilaterally flattened, 11th ovoid. Apical organs: well defined, small, nearly regular fossae located at anterior margins of 8–10th, on internal side of 4–7th, and on both sides of 11th; containing B4a sensillae with very long apical filaments (Fig. 151), B4b, M5, and C7 types. Lateral organs: small subapical zones of short and long B4c sensillae; no shortened uniporous sensillae have been found on microphotographs and it is unclear whether they are present or absent, which makes the affinity of *Aglaostola* to the following genera problematic. U1 sensillae occur at anterior margins, and on top of 11th; long, sparse A3 sensillae dispersed over glabrous surface.

Spectralia, *Hilarotes* (Figs 152–153, 155). Antennomeres 4–10 serrate-truncate, bilaterally flattened, 11th ovoid or irregular. Apical organs: well defined, large, transversely elliptical fossae located on internal side of 4–10th and on both sides of 11th; containing B4a sensillae with long apical filaments, M5 and C7 types; lateral organs: extensive fields of short U2 and long B4c sensillae. A3 sensillae dispersed over glabrous areas. *Ditriaena* (Fig. 154) differs from *Spectralia* and *Hilarotes* in having nearly regularly rhomboid antennomeres with straight inner margins, small apical fossae, and well defined sensillary fields comprising very dense, long B4c and short U2 sensillae.

Pterobothris (Figs 156–159). Antennomeres 5–10 transverse triangular with inner margins truncate, poorly bilaterally flattened, 11th irregular. Apical organs: small, deep, circular fossae on anterior surface of 5–11th (Fig. 156); containing B4a sensillae with very long apical filaments, M5 and C7 types (Fig. 157). Lateral organs: well defined, externally shifted depressions at inner margins (Fig. 158), containing long B4c and shortened U1 sensillae (Fig. 159, B4c, U1); long A3 sensillae dispersed over glabrous areas (Figs 156, 158).

Neobubastes, *Eububastes* (Fig. 166). Antennomeres 4–10 elongate triangular, slightly bilaterally flattened; 11th irregular. Apical organs: small, nearly circular fossae on anterior surface slightly, on proximal antennomeres more distinctly, internally shifted, 11th bearing circular fossa apically; containing B4a, M5, and C7 sensillae; lateral organs: poorly defined fields of B4c and shortened U1 sensillae along inner margins. Antennomere surface mainly glabrous, with U1 sensillae at anterior corners and on top of 11th, and long scattered A3 sensillae.

Thomassetia, *Augrabies*, *Jakovleviola* (Figs 168–170). Antennomeres 4–10 from strongly elongated, slightly broadened towards apex and weakly flattened (*Thomassetia*, Fig. 168) to serrate-truncate, strongly flattened (*Jakovleviola*, Fig. 170); 11th subcylindrical to broadened, irregular. Apical organs: small, nearly circular fossae on anterior surface, slightly (more distinctly on proximal antennomeres) internally shifted; in *Jakovleviola* (Fig. 170) fossae located on internal side of all the distal antennomeres; 11th bearing circular fossa apically or subapically; containing B4a, M5, and C7 sensillae; lateral organs: poorly defined fields of B4c and shortened U1 sensillae along inner margins. Antennomere surface mainly glabrous, bearing long, scattered A3 sensillae; U1 sensillae occur at anterior corners and on top of 11th.

Aristosoma (Fig. 172). Antennomeres 4–10 elongate triangular, with slightly arcuate inner margins, bilaterally flattened. Apical organs: small, nearly circular subapical fossae at anterior margins, containing B4a, M5, and C7 sensillae; lateral organs: poorly defined fields of B4c sensillae near apical corners. Antennomere surface mainly glabrous, with long U1 sensillae at anterior corners and on top of 11th, and long, sparse, scattered A3 sensillae.

Philanthaxia (Fig. 173) has generally the same structure as *Aristosoma* (Fig. 172). It can be distinguished by its far larger apical fossae.

Kurosawaia (Fig. 174) also has similar antennal structure, differing in antennomeres serrated from 3rd antennomere, inner margins strongly arcuate, and external U1 and B4c sensillae arranging along entire margin.

Kisanthobia (Fig. 171). Antennomeres 4–10 strongly elongated, slightly broadened apically, poorly bilaterally flattened, 11th irregularly ovoid. Apical organs: small, transverse, regular fossae on anterior surface, containing B4a, M5, and C7 sensillae; lateral organs: poorly defined fields of B4c and relatively long U1 sensillae along inner margins. Antennomere surface mainly glabrous, bearing long U1 sensillae around apical fossae, along inner margins, and on top of 11th, and long, sparse A3 sensillae.

Trigonogenium (Figs 164, 165). Antennomeres 4–10 weakly elongate triangular, slightly bilaterally flattened; 11th irregular. Apical organs: very large, deep, internally shifted, nearly circular fossae at anterior margins, 11th bearing circular fossae on both sides; containing B4a, B4b, M5, and C7 sensillae; lateral organs: indefinite fields of sparse coeloconic B4c sensillae. Antennomere surface mainly glabrous, bearing U1 sensillae at anterior corners and on top of 11th, and long scattered A3 sensillae.

Coomaniella (Fig. 163). Antennomeres 6–10 transverse to elongate triangular, sometimes transversely flattened; 11th irregular. Dimorphism: male antennae of some species much longer than in females; distal antennomeres of male of *C. biformis* bear long brush-like pilosity (BÍLÝ & KALASHIAN, 1994). Apical organs: large, nearly circular fossae on anterior surface; 11th with large circular fossa internally; containing B4b, B4c, and M5 sensillae; lateral organs absent. Antennomere surface mainly glabrous, bearing long B4c and shortened U1 sensillae near apical corners which do not form defined zones, and long scattered A3 sensillae.

Curis, *Neocuropsis* (Fig. 167). Antennomeres 4–10 elongate triangular, nearly serrate-truncate, poorly bilaterally flattened, 11th ovoid. Apical organs: small, internally shifted, slightly transverse or nearly circular, regular fossae on anterior margins, containing B4a, M5, and C7 sensillae; lateral organs: poorly defined fields of B4c and short U1 sensillae along inner margins. Antennomere surface mainly glabrous, bearing U1 sensillae at apical corners and on top of 11th, and long sparse A3 sensillae.

Neocuris (Fig. 177). Antennomeres 5–10 transverse triangular with rounded apical corners, poorly bilaterally and transversely flattened, 11th ovoid. Apical organs: regular, transverse, slightly compressed fossae at the middle of anterior margins, containing B4a, M5, and C7 sensillae; lateral organs: poorly defined zones of long and short B4c, and shortened U1 sensillae concentrated at apical corners; remaining surface glabrous bearing scattered, sparse A3 sensillae.

Torresita (Fig. 175). Antennomeres 5–10 elongated to elongate triangular, nearly serrate-truncate, poorly bilaterally flattened, 11th ovoid. Apical organs: small, transverse or nearly circular, regular, internally shifted fossae on anterior margins, containing B4a, M5, and C7 sensillae; lateral organs: poorly defined fields of B4c and short U1 sensillae along inner margins. Surface mainly glabrous, bearing shortened U1 sensillae at apical corners and on top of 11th, and long sparse A3 sensillae. *Pseudanilara* has generally the same structure but its apical organs exhibit partly closed sensory cavities.

Theryaxia (Fig. 176). Antennomeres 3–10 strongly elongated, slightly broadened towards apices, poorly bilaterally flattened, 11th strongly elongated, subcylindrical. Apical organs: transversely compressed fossae distally becoming almost closed cavities, located on anterior margins and containing B4b, M5, and C7 sensillae; lateral organs: poorly defined fields of long B4c and short U1 sensillae along inner margins. Antennomere surface mainly glabrous, bearing shortened U1 sensillae at apices, as well as long A3 type.

Anilara (Fig. 182). Antennomeres 4–10 serrate-truncate, bilaterally flattened. Apical organs: transversely reniform, deep fossae at the middle of anterior margins, containing B4a, M5, and C7 sensillae; lateral organs: poorly defined fields of long B4c and shortened U1 sensillae along entire inner margins; remaining surface glabrous, bearing long A3 sensillae concentrated along outer margins.

Anthaxoschema (Fig. 183) differs in having completely closed apical cavities.

Notographus (Fig. 179) has strongly elongated antennomeres with lateral sensory fields located on the anterior half of flattened inner margins; apical cavities completely closed.

Melobasis (Figs 180–181), *Montrouzieretta*. Antennomeres 4–10 serrate-truncate, bilaterally flattened, 11th elongate, often irregularly ovoid. Apical organs: partly opened cavities at the middle of anterior margins, containing B4a, M5, and C7 sensillae (Fig. 181); lateral organs: extensive fields of long B4c and shortened U1 sensillae along entire inner margins (Fig. 180); remaining surface glabrous, bearing scattered, long A3 sensillae.

Cylindrophora, *Paracuris* (Figs 184–185); see also Cobos (1956a: Figs 14–17, 23). Antennomeres 4–10 serrate-truncate, bilaterally flattened, 11th irregular or ovoid. Apical organs: open (*C. maulica*, *C. verecunda*) or completely closed cavities in the middle of anterior margins; according to Cobos (1956a; Fig. 15) *C. verecunda* has large apical fossae but I found open cavities instead; containing B4a, M5, and C7 sensillae; lateral organs: extensive fields of long B4c and shortened U1 sensillae along entire inner margins; remaining surface glabrous, bearing scattered A3 sensillae.

Tetragonoschema (Fig. 178). Differs in having short triangular antennomeres and lateral fields located beneath apical margins much resembling *Neocuris* (Fig. 177), but its apical cavities are almost completely closed.

Chalcogenia, *Anthaxia*, *Cratomerus*, *Brachelytrium*, *Brachanthaxia* (Figs 186–190); see also VOLKOVITSH & BÍLÝ (1997; Figs 25–30). Antennomeres 4–10 vary in shape from subcylindrical and serrate-truncate to transverse triangular, rhomboid, or leaf-shaped, bilaterally flattened, sometimes strongly enlarged (some *Cratomerus*); 11th also varies in shape. Dimorphism: coloration and size of distal antennomeres often different in males and females. Apical organs: regular fossae of variable shape at the middle of anterior margins of 4–10th and at the apex of 11th; containing B4a, M5, and C7 sensillae; lateral organs: extensive fields of B4c and short U2 sensillae along inner margins. Antennomere surface mainly glabrous, shortened uniporous sensillae occur at anterior corners and on top of 11th, A3 sensillae short, dispersed over the surface; sometimes coeloconic B4c sensillae dispersed on the sides (*Chalcogenia*, Fig. 186) as well. *Brachanthaxia* (Fig. 190) has apical fossae covered from above with a lobe, which is an extended, long projection of the inner margin of the fossa itself; this projection dips into the fossae along its entire length at antennomeres 9–10.

Xenorhipis, *Hesperorhipis* (Figs 191–193). Antennal structures demonstrate an extreme sexual dimorphism: in males, antennae pectinate from 2nd or 3rd with antennomeres densely covered with highly specialised bifurcate S8 sensillae (Fig. 192); in females these are serrate from 4th. Apical organs: poorly defined fossae located at the base of lateral projections that are flattened in cross section (male, Fig. 191), or on the sloping anterior surface (female, Fig. 193); containing B4a, M5, and C7 sensillae. Lateral organs indefinite: in female coeloconic B4c sensillae occur next to apical organs, short ?U2 sensillae situated on apical corners; in male B4c sensillae situated on the tops of lateral projections. Long A3 sensillae scattered over glabrous surface.

Trichinorhipis (Figs 194–195). Antennal structures also show sharp dimorphism: in males, antennae pectinate and covered with long capillaceous S8 sensillae (Fig. 194), in females these are serrate from 4th (Fig. 195). Apical and lateral organs not found (?absent). In the male, upper surface of lateral projections of antennomeres that are round in cross section bears sparse ?B4c sensillae. Antennomeres of female strongly elongated, weakly thickened distally, bear ?B4c sensillae on anterior corners and long A3 sensillae over entire surface (Fig. 195).

Juniperella (Figs 196–197). Antennomeres 3–10 strongly elongated, serrate-truncate, slightly broadened distally, poorly bilaterally flattened; 11th subcylindrical. Apical organs: open cavities at anterior margins of 3–10th and on top of 11th; containing B4a, M5, and C7 sensillae; lateral organs: extensive fields of coeloconic B4c and shortened U1 sensillae along entire inner margins, longer and denser at apical corners (Fig. 197) and on top of 11th; remaining surface glabrous, bearing sparse, scattered A3 sensillae.

Xenomelanophila, *Phaenops*, *Melanophila*, *Trachypteris* (Figs 198–201). Antennomeres 3(4)–10 strongly elongated, elongate triangular or serrate-truncate, poorly bilaterally flattened; 11th elongated, usually irregularly ovoid. Apical organs: partly or completely closed cavities at anterior margins of 4–10th and on top of 11th; only B4a sensillae are visible inside the open cavities; lateral organs: extensive, well defined fields of sparse B4c and dense U1 and U2 sensillae along inner margins, forming two zones (Figs 198, 200) of which anterior zone contains longer U1 sensillae, and posterior one very short U2 sensillae; remaining surface glabrous, bearing sparse A3 sensillae. *Melanophila* differs in having apical cavities completely closed (Fig. 201, AC) and only short U2 sensillae which are barely longer anteriorly.

Merimna (Fig. 202). Antennomeres 4–10 serrate-truncate, bilaterally flattened; 11th elongate ovoid. Apical organs: completely closed cavities at anterior margins of 4–10th and on top of 11th; no internal sensillae visible; lateral organs: fields of very short coeloconic B4c and U2 sensillae along inner margins, the latter usually forming groups of several sensillae arranged linearly. Surface mainly glabrous, bearing sparse A3 sensillae.

Cromophila (Figs 207–208). Antennomeres 4–10 serrate-truncate, bilaterally flattened; 11th ovoid. Dimorphism: antennomeres in males enlarged and differently coloured from those in females (Cobos 1986a,b). Apical organs: deep, internally shifted on proximal antennomeres, nearly circular fossae on anterior margins of 4–10th and near the apex of 11th; containing B4a, B4b, M5, and C7 sensillae. Lateral organs: irregular depressions or fossae on internal face also containing internal sensillae and often adjacent to apical ones on proximal antennomeres, and fields of sparse B4c and dense U2 sensillae along inner margins. Antennomere surface mainly glabrous, bearing sparse, long, scattered A3 sensillae.

Pseudactenodes, *Belionota*, *Actenodes* (Figs 209–212). Antennae serrate (*Pseudactenodes*: Fig. 212) to geniculate with 3rd antennomere much longer than 2nd (in *Pseudactenodes* 3rd weakly elongated); distal antennomeres sometimes asymmetrical, attaching at an angle to transverse antennal axes. Antennomeres 4–10 vary in shape (particularly in *Actenodes*), serrate-truncate, sometimes strongly elongated (Fig. 211) or transverse, bilaterally flattened; 11th usually of irregular shape. Dimorphism: antennomeres occasionally enlarged and bearing specialised S8 sensillae (Fig. 211) in males, sometimes the disposition and shape of sensory organs also differ in each sex. Apical organs: fossae of variable shape on internal side, sometimes strongly compressed but always open, often becoming confluent with lateral ones (Fig. 209), sometimes forming long furrows (Figs 210, 211), or transformed into deep cavities opening to the outside with narrow slits (*Pseudactenodes*, Fig. 212); containing B4a, M5, and C7 sensillae; 11th usually bearing two separated fossae (Fig. 211). Lateral organs: zones, fields, depressions or pits containing B4c sensillae, located on both sides, often becoming confluent with apical fossae or separated from them by thin septa (as in Agaeocerina); when antennomeres are asymmetrical the external fields shift to inner margins (as in Chrysobothrini). U1 sensillae (if present) are sparse, long, occurring along inner margins and at the top of 11th (Figs 209–210), remaining surface mainly glabrous, bearing short, scattered A3 sensillae.

Chrysobothris, *Colobogaster*, *Afrobothris* (Figs 203–206). Antennae geniculate with 3rd antennomere much longer than 2nd, distal antennomeres sometimes serrate-truncate, usually transverse, asymmetrical, attaching at an angle to transverse antennal axes. Antennomeres 4–10 vary in shape, usually transverse serrate-truncate, bilaterally and transversely flattened; 11th elongate ovoid, often irregular. Dimorphism: antennomeres occasionally bear long, capillaceous, specialised S8 sensillae along inner margins (Fig. 206) in males. Apical organs: always partly or completely closed cavities at anterior margins of 4–10th and on top of 11th; internal sensillae usually hidden within cavities. Lateral organs: fields of B4c and short U2 sensillae on both sides; when antennomeres are asymmetrical the lateral fields shift to expanded inner margins. Shortened U1 sensillae occur near apical organs (Figs 203, 205); in *Afrobothris* (Fig. 206) lateral fields consist of very dense coeloconic basiconic sensillae on internal and partly on external side; remaining surface glabrous, bearing scattered A3 sensillae.

Fahraeusia (Fig. 216). Antennomeres 4–10 elongate triangular, bilaterally flattened, 11th ovoid. Apical organs: well defined, medium-sized, circular, internally shifted fossae on anterior surface, 11th bearing circular fossa subapically; sensillary composition not studied because the fossae were completely filled with wax. Lateral organs: poorly defined small zones of external styloconic B4c sensillae on both sides. Antennomere surface mainly glabrous, with dense U1 sensillae along inner margins and on top of 11th, and long scattered A3 sensillae.

Galbelloid complex

Galbella (Figs 217–220). Antennomeres: 11; 5–10 serrate or pectinate, transverse, poorly bilaterally and strongly transversely flattened; 11th transverse, irregular. Dimorphism remains to be researched; the single studied *G. felix* specimen, of unknown sex, bears dense, long, curved basiconic sensillae distally which may or may not be of a special male type (Figs 217–219, ?S8); such sensillae (Fig. 219, B4b, M5) have not been found in *Galbella* sp. (Fig. 220). Apical organs: poorly defined irregular depressions on anterior surface, containing long semi-erect B4b and solitary M5 sensillae sometimes occurring outside depressions; no C7 sensillae were found. Sensory zones on apical corners containing sparse, long basiconic (?B4c) and shortened U1 sensillae (Fig. 220), sparse A3 sensillae scattered over the surface.

IV. Evolution and general trends in the transformation of antennal structures in Buprestidae

Schizopoid complex (Figs 1–4)

The composition, structure and arrangement of sensillae and sensory organs in the Schizopoid complex are in complete agreement with those in other Buprestidae except that external B4c and A3 sensillae are widely dispersed over the surface, which is a primitive state. The significant variability of antennomeres in size, shape and number within the same species or even sex (NELSON & BELLAMY 1991) confirms that schizopodine antennae are relatively less specialised. Two phyletic lineages can be recognised within this complex. Schizopodini (Fig. 1) undoubtedly exhibit the most primitive state (11 antennomeres, poorly defined apical depressions) whereas Dystaxiini (Figs 2–4) display more advanced states (12 antennomeres, relatively defined subapical depressions or fossae) corresponding to chalcophoroid (*Dystaxia*, Fig. 4) or buprestoid (*Glyptoscelimorpha*, Fig. 2) pathways of sensory organ genesis. The main evolutionary trends within the Schizopoid complex are the differentiation of apical organs, an increase in the number of antennomeres, and development of sexual dimorphism.

Julodoid complex (Figs 5–9)

The sensory structures are rather uniform in all the studied taxa of this complex. Although sensillary composition and structure are generally the same as in other higher buprestid taxa, modified A3 sensillae, extensive sensory fields, and very peculiar sensory zones are unique to this complex. Julodinae show high differentiation of sensillae and low differentiation of the sensory organs; the latter are even more primitive than those of the Schizopodinae. The main evolutionary trend within this complex is the development of lateral fields, followed by strong bilateral flattening of the antennomeres.

Polycestoid complex (Figs 10–43)

The sensory organs in the Polycestoid complex are formed by concentration of internal sensillae in superficial zones near antennomere apices, followed by their subsequent transformation into depressions and fossae, sometimes combined with differentiation of lateral fields. The general Polycestoid morphocline includes the following stages: 1) appearance of apical zones or poorly defined depressions made up of internal sensillae and surrounded with external B4c sensillae (Acmaeoderioid lineage; Figs 10–20); 2) transformation of apical depressions into pits and fossae sometimes followed by incorporation of external B4c sensillae into lateral or apical organs (Polycestioid lineage; Figs 21–31); 3) and the differentiation of lateral fields made up of external sensillae (Polyctesioid lineage, Prospherini, *Astraeus*; Figs 32–40, 42–43). In general, the transformation of sensory organs in this complex has much in common with that in Buprestinae, being restricted to more primitive states.

Acmaeoderioid lineage (Mastogeniini, *Odettea*, Ptosimini, Nothomorphini, *Acmaeoderoides*, Acmaeoderini, Paratrachyini) (Figs 10–20). Sensory organ differentiation is restricted to the 1st stage, as described above, revealing the most primitive states in this complex. The more advanced states are characteristic of Paratrachyini (Figs 19–20). Another trend that is inherent in the Acmaeoderioid lineage is the development of marked sexual dimorphism characterised by enlarged male antennomeres, resulting in extension of their surface and often followed by dispersion of dense basiconic (Acmaeoderini) or trichoid (Mastogeniini) sensillae. Mastogeniini (Figs 10–11), showing some distinctions in apical organ structure and manifestation of dimorphism, possibly belong to their own phyletic lineage.

Polyctesioid lineage (Thrincopygini, *Chrysophana*, *Beerellus*, Polyctesini) (Figs 32–40) is characterised by the occurrence and further differentiation of lateral fields of external B4c and shortened U1 sensillae (the 3rd stage) followed by lengthening and flattening of antennomeres and shifting of the apical fossae towards the internal face.

Sexual dimorphism occurs, characterised by moderate enlargement and lengthening of male antennomeres bearing denser basiconic sensillae. The sensory formations of Thrincopygini (Figs 32–34) (undifferentiated subapical zones and lateral fields) are generally primitive, although the areas of very dense A3 sensillae in the apical part of antennomeres, resembling the lateral fields of Julodini, are inherent in this group – evidence of specialisation. This means that Thrincopygini possibly represent their own phyletic lineage. *Chrysophana* (Figs 40) also demonstrates a special state (lateral fields of coeloconic B4c sensillae). Among Polyctesini the most primitive state, which is similar in many respects to that of Ptosimini and Acmaeoderini, is found in *Schoutendeiastes* (Fig. 35); the most advanced state occurs in *Polyctesis* (Figs 38–39). All the intermediate states can be traced in succession: *Schoutendeiastes – Bellamyina – Paraxenopsis – Polyctesis* (Figs 35–39). In general, sensory organ genesis within Polyctesioid lineage follows the buprestoid pathway.

Polycestioid lineage (*Bulis*, Polycestini, Tylauchenini, Tyndarini, Acherusini, Pseudacherusini) (Figs 21–31). Specialisation in this lineage leads towards the formation of well defined regular apical fossae (the 2nd stage). Additional, often confluent apical and lateral fossae that have no fixed position sometimes appear in intermediate stages (Figs 25, 27). The differentiation and deepening of the fossae is usually followed by the unification of B4 sensillae. Sexual dimorphism is not generally marked. Sensory organ transformation can be traced in succession: *Bulis* (Fig. 21) (apical fossae poorly differentiated, B4c and A3 sensillae dispersed over the surface) – Polycestini (Figs 22–27) (deep, variable in shape and position apical and lateral, or only apical fossae) – Tylauchenini, Tyndarini, Acherusini, and Pseudacherusini (Figs 28–31) (proximal antennomeres lack fossae). It may be suggested that Polycestini (the more primitive group) and Tylauchenini-Pseudacherusini (the most advanced groups) form two closely related phyletic branches. Within Polycestini the external B4c sensillae initially concentrate into additional depressions or fossae (*Strigopteroides*, *Strigoptera*, *Jelinekia*, *Polycesta*) (Figs 22, 26–27) which subsequently combine with apical fossae or disappear as a result of unification of sensillae of B4 type (*Paracastalia*, *Polycestina*, *Pseudocastalia*, and *Thurntaxisia*) (Figs 23–25). The same trends (apical organ differentiation followed by size reduction and 4Bc sensillae incorporation into fossae) are characteristic of the second branch but no additional fossae can be traced, while apical organs disappear on proximal antennomeres.

Finally, the Polycestoid complex includes three enigmatic Australian groups: Xyroscelini, Prospherini, and Astraeusini. Their morphocline direction is similar to that in Polyctesioid lineage. *Xyroscelis* (Fig. 41) shows the most primitive state (poorly defined apical depressions, lateral fields nearly absent). In *Prospheres* (Fig. 43) and *Euleptodema* the further differentiation of apical fossae and the formation of extensive, poorly defined lateral fields of B4c sensillae can be observed; this state has much in common with those observed in some Buprestoid taxa (Figs 121–128, 131–134). *Astraeus* (Fig. 42) demonstrates the most advanced state (well developed subapical fossae and lateral fields deepened into shallow depressions).

Chalcophoroid complex

Sensory organs in Chalcophoroid complex originate in the confluence of primarily single, widely dispersed pores containing coeloconic sensillae into larger organs (chalcophoroid pathway of genesis). Different trends occur in different phyletic lineages and branches, but the general chalcophorine morphocline is made up of the following stages: 1) confluence of the single pores into irregular depressions and small pits having no fixed position (*Vadonaxia*, *Paratassa*, *Nanularia*, *Ampheremus*, *Chalcoplia*, *Descarpentriesiola*, *Embrikillium*, Chrysochroini – Chalcophorini, *Euchroma*, *Hypoprasis*, *Baudonisia*, Pseudoperotina) (Figs 44–67, 73–74, 101–102); 2) further confluence and differentiation of depressions and pits giving rise to large fixed organs of which the apical ones are the first to form (*Halecia – Eupodalecia – Euplectalecia – Cinyra*; *Hippomelas – Achardella – Pseudolampetis – Ectinogonia*, *Chalcopoecila*; Sphenopterini; *Polybothris sulcicollis – Capnodis – Perotis – Lampetis*) (Figs 75–81, 89–100, 103–109); 3) oligomerization of sensory organs, presumably resulting from the unification of B4-type sensillae, leads to the formation of large, single apical organs on anterior surface or internal face followed by the transformation of antennomeres from elongated to triangular (*Saundersina*, Poecilonotini, *Dicerca*, *Dicercomorpha*, Haplotrinchina) (Figs 82–88, 111–115). Sometimes the genesis of sensory organs follows the path of modification when the initial stages of a morphocline can hardly be traced, as occurs in Evidini (Figs 68–72). Depending on the presence or absence of fields or zones of U2 sensillae , two general lineages may be recognised within Chalcophoroid complex.

Chalcophorioid lineage. U2 sensillae lacking; only *Chalcophora* and some related taxa have zones of shortened U1 sensillae (Fig. 65, U1).

The most primitive states are found in *Vadonaxia*, *Paratassa*, *Nanularia*, and *Ampheremus* (Figs 44–47). The sensory organs of *Vadonaxia* (Fig. 44) are presumably of the most archaic type, resembling those in primitive Polycestioid taxa (poorly defined, frequently confluent depressions of variable shape and disposition, well distinguished, mostly superficial basiconic sensillae). A close affinity between *Paratassa* to *Nanularia* and *Ampheremus* (Fig. 45–47), is extremely doubtful, in spite of quite similar antennal structures; these probably result from symplesiomorphy.

The sensory organs of *Chalcoplia*, *Descarpentriesiola*, and *Embrikillium* (Figs 48–51) can also be regarded as primitive, especially those of *Chalcoplia* (Fig. 48) and *Descarpentriesiola* (Figs 49–50) (schizopoid antennomeres with poorly defined apical depressions on anterior surface, lateral fields of single and confluent pores and small depressions). *Embrikillium* (Fig. 51) exhibits the more advanced state (antennomeres elongated and more flattened with better defined apical and lateral depressions), bearing some similarity to those of Neotropical chalcophorine taxa and Psilopterini. *Descarpentriesiola* and *Embrikillium* (Figs 49–51) bear dense, shortened U1 sensillae apically. This group exhibits its own morphocline, which partly coincides with that of Chalcophorini and Psilopterini, but is confined to the initial stages of the general Chalcophoroid morphocline.

Two close morphoclines that are primarily determined by variability of antennomere shape can be traced within the Chrysochroini-Chalcophorini group. Chrysochroini (Figs

52–54) is characterised by the transverse extension of distal antennomeres and their shifting around transverse axes, which gives rise to pectinate or flabellate antennae bearing pores or small pits dispersed over the surface or sometimes becoming confluent to form larger fossae, with the most advanced state in *Agelia* (Fig. 53). In contrast, in Chalcophorini (Figs 55–66) the antennomeres are lengthened and bilaterally flattened, followed by the differentiation of lateral fields of pores and small pits sometimes also becoming confluent to form larger fossae, with the most advanced state in *Chalcophora* (Figs 65–66). Both morphoclines are characterised by a lack of the topographically fixed fossae that can arise occasionally as a result of confluence of pores and small pits on different parts of the antennomeres. In spite of sharp differences between the most advanced states, it is impossible to make a distinct delimitation between Chrysochroini and Chalcophorini based on antennal organs. It may be concluded that the state of *Cyphogastra* (Fig. 61) and closely related genera (antennomeres moderately elongated and poorly bilaterally flattened, pores and small pits uniformly dispersed over the surface, occasionally confluent into larger apical and/or lateral depressions or fossae) is initial and the most primitive for both morphoclines. Quite similar states and trends can be traced in the intermediate stages, making it difficult to attribute some genera to a certain group. For example, *Chalcophorella* (Fig. 60) and *Stigmatophorella* on the one hand, and *Nipponobuprestis* (Fig. 62) and *Texania* on the other, demonstrate very similar transverse apical depressions; however, on the basis of antennomere shape, these taxa most likely belong to different generic groups. The confluence of pores and pits into extensive lateral depressions is a characteristic of the *Sapaia* group (Figs 56–58) on the one hand, and *Scaptelytra* (Fig. 59) on the other. In antennal structure (sharply triangular antennomeres, sensory depressions at the centre of anterior surface, and lateral fields well defined) only *Chlorophorella* (Fig. 67) differs greatly from other taxa, which points to its isolated position.

The extremely modified antennal organs of *Evides* (Figs 68–72) presumably arise from initial states similar to those in representatives of *Sapaia* group (Figs 56–58), in which the proximal antennomeres bear very extensive depressions (Fig. 58). Their further differentiation, followed by the lengthening and partial unification of internal sensillae (Fig. 72), leads to the formation of unusually large organs that can invaginate inwards, making the antennomeres asymmetrical (*E. kraatzi*, Fig. 69).

A peculiar morphocline is inherent in Neotropical chalcophorine genera. Its initial states correspond almost completely to those in Chrysochroini (*Euchroma*, Fig. 73) or Chalcophorini (*Hypoprasis*, *Baudonisia*, *Cordillerita*; Fig. 74), but in the latter stages pores and pits become confluent to form two large, irregular, sometimes joined subapical and lateral depressions internally, and only lateral ones externally, as can be observed in *Halecia* and *Eupodalecia* (Figs 75–76). The depressions change into regular circular fossae, of which the subapical ones differentiate earlier (*Euplectalecia*; Figs 78–79) than the lateral (*Cinyra*; Figs 80–81). The most advanced state (very large, regular subapical fossae on the internal face) is found in *Saundersina* (Fig. 82), in which organs are presumably formed either by confluence of subapical and lateral fossae, or disappearance of lateral fossae. The disposition of subapical and lateral fossae in *Chrysesthes* (Fig. 77) and *Pelecopselaphus* is in complete agreement with that in *Euplectalecia* and *Cinyra* (Figs 78–81), whereas their differentiation level corresponds to *Euplectalecia* (Figs 78–79), but these genera differ in having strongly transverse antennomeres. In such a manner, despite the fact that sensory organ transformation in this branch follows the same path as in Chrysochroini – Chalcophorini, the most advanced states are in full agreement to those in Psilopterini: *Cinyra* state corresponds to states in *Ectinogonia*, *Chalcopoecila*, and some *Lampetis* (Figs 98–100, 108), and *Saundersina* state is in agreement with that in *Dicerca* (Fig. 111). *Chrysesthes* and *Pelecopselaphus* form their own group, which combines the characters of both *Euchroma* and *Hypoprasis*-*Cinyra*; their external similarity to *Agaeocera* (Figs 137–138) from Buprestoid complex, bearing only internal fossae, results in all probability from parallel evolution.

Palmar, *Poecilonota*, and *Nesotrinchus* (Figs 83–88), having glabrous schizopoid antennomeres with very large subapical depressions or fossae, also demonstrate a specific morphocline. Their sensory organs probably formed by concentration of coeloconic sensillae at the anterior part of antennomeres followed by their confluence into single large apical organs, as can be observed in the succession: *Palmar* (*Scintillatrix*) *limbata* (Figs 83–84) – *P.* (*S.*) *chinganensis* (Fig. 85) – *P.* (s.str.) *virgata*, *Poecilonota*, *Nesotrinchus* (Figs 86–88). The similarity of the antennal structures to those in *Saundersina* (Fig. 82), *Dicerca* (Fig. 111), *Dicercomorpha*, *Cardiaspis*, *Haplotrinchus* (Figs 113–115), and some Buprestoid taxa may also be a result of parallel evolution.

Psilopterioid lineage. Antennomeres with fields or groups of short U2 sensillae along inner margins.

Sphenopterini (Figs 89–94) can be included in Psilopterioid lineage because of the presence of U2 sensillae, which do not form defined fields, along inner margins (Fig. 94, U2). The genesis of sensory organs in this group follows the path of the formation of regular apical fossae (Fig. 94, AF) and poor differentiation of lateral zones or fields of mainly separate coeloconic sensillae (pores) (Fig. 94, LF). A distinct morphocline from the most primitive states (antennomeres of schizopoid type, apical depressions poorly defined, lateral zones indefinite) to the advanced (antennomeres elongated and strongly flattened, apical fossae regular, deep, internally shifted; lateral fields extensive) can be traced in the following succession: *Sphenoptera* (*Sphenopterella*) (Fig. 92) – *Genestia* (Fig. 89) – *Sphenoptera* (s.str.) (Fig. 93) – *Evagora* (Fig. 91), *S.* (*Chrysoblemma*) (Fig. 94), *Armenosoma* (Fig. 90). Hence, with reference to sensory organ states, Sphenopterini is the most primitive separate branch of Psilopterioid lineage.

In another branch of this lineage, Psilopterini (Figs 95–115), U2 sensillae form well defined zones and fields (Fig. 95, U2). The morphocline of sensory organ states consists of the following stages: 1) dispersion of single and confluent pores or pits over anterior surface and both sides of antennomeres (*Asidoptera*, *Pseudoperotis*; Figs 101–102); 2) appearence of apical depressions at anterior surface (*Hippomelas*, *Achardella*, *Polybothris sulcicollis*; Figs 95, AD; 103); 3) differentiation of lateral fields of pores and small pits (*Capnodis*, *Cyphosoma*; Figs 105–106) or their confluence into a few or single fossae (*Ectinogonia*, *Chalcopoecila*, *Polybothris staudingeri*, *Perotis*, *Lampetis*, *Latipalpis*; Figs 97–100, 104, 107–109); 4) formation of a single subapical internal fossa

(*Dicerca*; Figs 111–112) or an apical one (*Dicercomorpha*, *Cardiaspis*, *Haplotrinchus*; Figs 113–115) as a result of further oligomerization followed by 4B sensillae unification and antennomere shape transformation.

The total agreement in at least three initial stages between New World and Old World psilopterine genera suggests that these genera form closely related groups that have presumably evolved independently in each part of the world. The huge and very probably heterogenous Madagascan *Polybothris* – *Icarina* Alluaud, 1896 conglomeration, which demonstrates both primitive (*P. sulcicollis* resembling *Hippomelas*; Figs 95, 103) and advanced (*P. staudingeri* resembling *Ectinogonia* and *Lampetis*; Figs 98, 104, 108) states, apparently also form their own group. *Asidoptera* and *Pseudoperotis* (Figs 101–102), possessing the most primitive organs among Psilopterini, can be regarded as one more separate group. The antennal structure of *Hippomelas* (Fig. 95), completely fitting with the primitive states of Psilopterini, testifies to their closest affinity, whereas the differences in proximal antennomere length (the 3rd is much longer than 2nd) suggest the separate position of this genus. The well defined single subapical fossae of *Dicerca* (Figs 111–112) very probably derived through the confluence of smaller pits, as can be seen from the proximal antennomeres of *Latipalpis* (Fig. 110). Their external resemblance to those in *Palmar* and *Poecilonota* (Figs 86–87) apparently results from convergence, because apical fossae arose in a different way (see above) in the latter, and their position, sensillar structure and composition, as well as the shape of the proximal antennomeres, differ greatly from *Dicerca*. Sensory organs of *Dicercomorpha*, *Cardiaspis*, and *Haplotrinchus* (Figs 113–115) demonstrate even greater similarity to *Palmar* and *Poecilonota* but these also have defined zones of U2 sensillae. Apical fossae formation in this group apparently follows the same path as Poecilonotini, that is the concentration of pores or small pits on the anterior surface followed by their complete confluence.

Buprestoid complex

Apical organs arise through the formation of depressions comprising internal sensillae near antennomere apices and their further transformation into fossae and then to open and closed cavities (buprestoid pathway of sensory organ genesis); lateral organs are usually fields of the external B4c and U1–U2 sensillae that sometimes deepen into well defined depressions and fossae (*Agaeocerina*, *Cromophila*, *Actenodini*; Figs 135–137, 207–211) or, rarely, are absent (*Nascionina*, *Coomaniellini*; Figs 160–163). The general Buprestoid morphocline consists of the following stages: 1) dispersion of single sensillae over the entire antennomere surface or concentration of internal sensillae into indefinite apical depressions (Mendizabalini, Maoraxiini; Figs 116–118, 213–215); 2) a differentiation of the apical depressions on the anterior surface, lateral fields absent (Nascionina, Coomaniellini; Figs 160–163); 3) a transformation of the apical depressions into regular fossae and the occurrence of extensive lateral fields, sometimes deepened into depressions or fossae made up of B4c sensillae; sometimes these fields are formed by predominantly U2 sensillae (Buprestioid lineage) (Figs 119–159); 4) a subsequent transformation of the apical fossae into cavities and further differentiation of the lateral fields involved into external basiconic and short uniporous sensillae (Anthaxioid lineage) (Figs 163–190); 5) a differentiation of the apical cavities (except *Cromophila* which bears regular fossae, Figs 207–208) and lateral organs accompanied by the transformation of antennae from serrate to geniculate and asymmetry of antennomeres (Chrysobothrioid lineage) (Figs 196–212).

The most primitive states are a lack of sensory organs (*Mendizabalia*, Figs 116–117) or their representation by poorly defined subapical depressions (*Philandia*, Fig. 118) that makes it impossible to attribute these taxa to any certain phyletic lineage.

The next state in specialisation is demonstrated by Maoraxiini (Figs 213–215) (indefinite subapical depressions, no lateral fields) in which the sensory formations to some extent resemble those in Mastogeniini (Figs 10–11) preventing their being placed in any phyletic lineage. Maoraxiini should probably be regarded as an isolated group at the base of Buprestoid phyletic stock.

Buprestioid lineage. This lineage is characterised by large, usually poorly defined subapical organs on the internal face and extensive fields of B4c sensillae, (U2 sensillae are also occasionally involved or may even predominate); in some groups the fields transform into defined zones, depressions, or fossae. With regard to the position and differentiation level of sensory organs, the structure of internal sensillae, and the composition of external ones, some separate branches and isolated groups may be recognised within this lineage.

The sensory organs of *Julodimorpha* (Fig. 119) closely resemble those in *Buprestis*, *Eurythyrea* (Figs 128–129), and other taxa belonging to the next branch but their inner structure of confluent fragments and some other features suggest that this genus is rather isolated.

The central branch of Buprestioid lineage consists of taxa with antennae bearing normal or slightly shortened external U1 sensillae. One of the most primitive states is found in *Pygicera* (Fig. 120) which, in spite of its well developed apical fossae, has no defined fields. *Nascioides* and *Nascio* (Figs 160–162) demonstrate poorly defined apical depressions or fossae on the anterior surface and undeveloped lateral fields. Differences in the shape and position of the apical organs between *Nascio* (Figs 161–162) and *Nascioides* (Fig. 160) cast some doubt upon the extreme closeness of their affinity. The apical organs of *Nascio*, which are better defined, regular, deep, markedly internally shifted, and bordered with additional depressions, bear some external similarity to those in Poecilonotini and especially to *Nesotrinchus* (Fig. 88). The antennal structures of *Nascioides* (Fig. 160), apart from being of very variable antennomere shape, are similar to those in *Coomaniella* (Fig. 163) and *Kisanthobia* (Fig. 171). Epistomentini (Figs 121–123), *Neobuprestis* and *Balthasarella* (Figs 124–126), *Trachykele* (Fig. 127), and *Lamprocheila* (Fig. 131) have poorly defined subapical depressions or fossae and extensive lateral fields of 4Bc sensillae. Phrixiini (Figs 132–133) differ in having smaller and better defined subapical fossae, and lateral fields made up of shortened uniporous sensillae as well. Further specialisation is mainly determined by lateral organ differentiation that can be traced in succession as follows: *Yamina* (Fig. 130) – *Buprestis* (Fig. 128) – *Eurythyrea* (Fig. 129) – *Mixochlorus* (Fig. 135) – *Peronaemis* (Fig. 136) –

Agaeocera (Fig. 137). The last shows the most advanced state of this morphocline, resembling those in *Pelecopselaphus* and *Chrysesthes* (Fig. 77) from Chalcophoroid complex, but in *Agaeocera* (Figs 137, 138) the lateral organs are located only on the internal face and originated in another fashion. A similar state which fits the mentioned morphocline is also found in *Exagistus* (Fig. 134) but its close affinities to Agaeocerina are extremely doubtful. One more enigmatic genus, *Fahraeusia* (Fig. 216), possessing well developed, small apical fossae, barely visible lateral zones of external styloconic sensillae, and very dense, long, uniporous ones apically that look like those in *Embrikillium* (Fig. 51), can also be attributed, extremely tentatively, to this lineage .

Within another branch of Buprestioid lineage that includes Australian taxa exclusively, the short U2 sensillae form sensory fields in combination with those of B4c type; well defined apical fossae show a tendency to shift on the sloping anterior surface, a characteristic of many Stigmoderini (Fig. 147); sensory fields small, shifted towards inner margins (Figs 143–147). *Buprestina*, *Euryspilus*, *Bubastes*, and *Microcastalia* (Figs 139–143) have apical fossae that are deepest at the base and most shallow distally. The lateral field differentiation in the succession *Buprestina – Euryspilus – Bubastes inconsistans – B. iridescens* (Figs 139–142) follows the same path as in the previous branch. The lateral fields of *Microcastalia* (Fig. 143) are distinctly shifted to inner margins, as occurs in Stigmoderini (Figs 144–147, 149). A feature peculiar to *Buprestina* (Fig. 139) is that internal B4a sensillae extend into very long apical filaments, and such sensillae are inherent in the next branch and *Pterobothris*. *Hypostigmodera* (Figs 148–149) stands out from other taxa because of its 12 antennomeres and sharp dimorphism.

It is presumed that American *Aglaostola*, *Spectralia*, *Ditriaena*, and *Hilarotes* (Figs 150–155), in which the internal B4a sensillae bear very long apical filaments (Fig. 151) much like *Buprestina* (Fig. 139), also form a separate branch of Buprestioid lineage. Similar sensillary structure may prove the relationships of these taxa, but on the other hand modified sensillae could occur independently in different groups. *Aglaostola* differs from the other genera in having triangular antennomeres and poorly developed sensory fields made up almost exclusively of B4c sensillae (Fig. 150). *Spectralia* and *Hilarotes* (Figs 152–153, 155), having trapezoid antennomeres, and *Ditriaena* (Fig. 154) with rhomboid ones, show well developed and externally shifted fields of mainly U2 sensillae that are closely similar to those in Psilopterini.

Of particular interest is Neotropical *Pterobothris* (Figs 156–159), in which the sensory organs combine the characters of the two previous groups. Small, deep apical fossae with filamentous B4a sensillae (Fig. 157) are similar to those in *Buprestina* and *Aglaostola-Hilarotes*, whereas the sensory zones of B4c and U2 sensillae on inner margins (Fig. 159) resemble those in *Microcastalia* and Stigmoderini; but these zones are much more defined and bear distinctly lengthened sensillae. Through its characteristic sensory organs *Pterobothris* presumably represents an isolated branch within Buprestioid lineage or even a separate lineage of Buprestoid complex.

Anthaxioid lineage. This lineage is characterised by well defined apical organs located on the anterior surface or, rarely, shifted to the internal face of antennomeres. The transformation from apical fossae to open and then closed cavities can easily be observed in different groups. Lateral organs (excepting *Coomaniella* which has no marked external sensillary areas) are rather uniform, being represented with fields of B4c, U2, rarely U1 sensillae along inner margins. The antennal structures of Xenorhipini demonstrate sharp sexual dimorphism. Several branches can be established within the lineage, based on trends in sensory organ genesis

The most primitive state (poorly defined apical fossae on the anterior surface, lateral organs absent) is found in Coomaniellini (Fig. 163), although the fact that fossae appear onwards from the 6th antennomere only and a number of other important characters (BÍLÝ, 1974) attest that this tribe is highly specialised. Antennal structures of Coomaniellini differ from those of other Anthaxioid taxa, to some extent resembling sensory organs of *Kisanthobia* as well as *Nascioides*.

The next specialisation states are the defined apical fossae internally shifted or, rarely, located at the centre of the anterior surface, and poorly defined lateral fields. Their morphocline may be traced in the following succession: *Trigonogenium* (Figs 164–165) – *Neobubastes* (Fig. 166), *Eububastes* – *Curis*, *Neocuropsis* (Fig. 167) – Thomassetiini (Figs 168–170) – *Aristosoma* (Fig. 172), *Philanthaxia* (Fig. 173) – *Kurosawaia* (Fig. 174). The sensory organs of *Trigonogenium*, because of their large, internally shifted apical fossae, extensive fields of B4c sensillae, and relatively long U1 ones, resemble those in *Tylauchenia* (Fig. 28) from Polycestoid complex and some Buprestioid taxa, supporting the separate position of this genus. The great similarity in sensory organs represented by small apical fossae, slightly internally shifted at least on proximal antennomeres, and fields that are only poorly defined can be found within Australian *Neobubastes*, *Eububastes*, *Curis*, and *Neocuropsis* (Figs 166–167). A similar structure is characteristic of Ethiopian Thomassetiini, although *Thomassetia* (Fig. 168) differs from *Augrabies* and *Jakovleviola* (Figs 169–170) in antennomere shape as well as the shape and disposition of apical fossae. *Aristosoma*, *Philanthaxia*, and *Kurosawaia* (Figs 172–174) demonstrate fossae that have shifted more internally. The last-mentioned is distinguished from the others in having antennomeres nearly serrate-truncate and well defined fields comprising long U1 sensillae along the entire inner margin. The opposite trend can be observed in *Kisanthobia* (Fig. 171), which has apical fossae located at the centre of anterior surface, as occurs in Anthaxiini (Fig. 187).

Further specialisation produces transformation of the apical fossae located on the anterior surface into open and closed cavities and sensory field differentiation associated with the uniporous sensillae shortening and increasing in number. These trends predominate in Anthaxinioid branch. The primitive state (apical fossae instead of cavities) is demonstrated by *Torresita*, *Neocuris*, *Anilara*, *Chalcogenia*, *Anthaxia*, *Cratomerus*, and *Brachelytrium* (Figs 175, 177, 182, 186–189); that of *Torresita* is presumably the most primitive (apical fossae slightly internally shifted on proximal antennomeres) resembling states in the previous lineage but distinguished by much more defined fields of shortened U1 sensillae. The most advanced states (apical cavities closed, sensory fields made up of mainly U2 sensillae) are inherent in *Tetragonoschema*, *Notographus*, *Anthaxoschema*, and *Paracuris* (Fig. 178, 179, 183, 185); the intermediate states are found in *Theryaxia*, *Pseudanilara*, *Melobasis*, *Montrouzieretta*, *Cylindrophora*, and *Brachanthaxia* (Figs 176, 180–181, 184, 190). It may be surmised that similar structures within this lineage originated in different ways. For example, the cavities in the succession *Torresita – Neocuris – Theryaxia – Tetragonoschema – Notographus* (Figs 175–179) develop as the result of both the fossal margins closing, followed by invagination of the entire organ within the antennomere body. In African *Brachanthaxia* (Fig. 190) the cavity originated in the extension of only the inner fossal margins into long lobes that cover the fossae from above; all the transformation stages can be traced from proximal to distal antennomeres of the same antenna (see also VOLKOVITSH & BÍLÝ, 1997: 258, Fig. 30). These examples show that apical cavity formation can proceed in parallel in different groups and that similar states do not necessarily mean direct relationships.

A distinctive specialisation trend associated with sharp sexual dimorphism is demonstrated by Xenorhipini (Figs 191–195). General Buprestoid structure is preserved only in females (Figs 193, 195) whereas the males (Figs 191–192, 194) possess pectinate antennae bearing an external similarity to those of *Mendizabalia* (Fig. 116) but differing in well defined apical organs (Fig. 191) and modified external sensillae (Figs 192, 194). Apparently, the peculiar antennae and specialised sensillae of the male are associated with perception of female sex pheromones; the male to female attraction in *Xenorhipis brendeli* has been described by WELLSO (1966). With reference to the shape and structure of antennomeres, apical organs, and male sensillae, *Xenorhipis* and *Hesperorhipis* (Figs 191–193) differ greatly from *Trichinorhipis* (Figs 194–195). It should be stressed that the far more conservative female organs look much more primitive than those in the previous lineage. Thus the close relationships of Xenorhipini to Anthaxiini merit further study.

Chrysobothrioid lineage. Specialisation in this lineage involves not only sensillae and sensory organs but entire antennae as well. External sensillae differentiation results in shortening of uniporous sensillae which, in combination with basiconic ones, form lateral zones and fields sometimes deepened into depressions or fossae; furthermore, this is associated with sexual dimorphism characterised by the appearance of extremely modified serrate and trichoid male sensillae in Actenodini and Chrysobothrini (Figs 206, 211). Sensory organ transformations follow two main pathways that presumably correspond to two phyletic lineages.

Melanophilinioid branch. Apical organ differentiation is limited to transformation from primitive, open cavities in *Juniperella* (Figs 196–197) to advanced, closed ones in *Melanophila* (Fig. 201) and *Merimna* (Fig. 202). Lateral organ differentiation is associated with delimitation of two zones of long and short external sensillae in *Xenomelanophila* (Fig. 198) and *Phaenops* (Fig. 200) or equal shortening of uniporous sensillae in *Melanophila* and *Merimna* (Figs 201–202); the most primitive state is also found in *Juniperella* (Fig. 196). These transformations do not involve entire antennae. The basic pathway of genesis coincides to a large extent with that of some Australian Anthaxioid taxa (Figs 178–181, 183–185). However, the wide-open apical cavities and, especially, the extensive undifferentiated lateral fields in *Juniperella* that bear a great resemblance to primitive states in Buprestini, suggest that Melanophilini could have diverged from Buprestioid ancestors; in which case the similarity to *Melobasis* and related groups would result from parallel evolution.

Chrysobothrinioid branch. The differentiation of antennal structures in general coincides with that in the previous branch but the transformations are more complicated and involve entire antennae. The most primitive state (antennae serrate with 3rd antennomere relatively short and apical fossae circular) is found in *Cromophila* (Figs 207–208). Within Actenodini the complete shifting of apical fossae to the internal face, frequently followed by their closing in, or even confluence with, lateral organs (Fig. 209) can be observed. Sometimes these states are similar to those in *Pelecopselaphus* group and Agaeocerina (some *Actenodes* species). In *Belionota* and *Pseudactenodes* (Figs 210–212) the strong lengthening and compression of fossae leads to the appearence of longitudinal grooves and open cavities. The antennae themselves transform from serrate with 3rd antennomere relatively short (*Pseudactenodes*) to geniculate with strongly elongated 3rd (many *Actenodes* and *Chrysobothris*). Some American *Actenodes* exhibit geniculate antennae with transverse distal antennomeres bearing markedly compressed but always open apical fossae and lateral fields or depressions shifted to inner margins. The last state is intermediate to that in Chrysobothrini (Figs 203–206) which have geniculate antennae, asymmetric antennomeres shifted around transverse axes, and apical organs completely closed. The trend for lateral fields to dip into depressions or fossae is also well developed within Actenodini and Chrysobothrini.

Galbelloid complex (Figs 217–220)

The antennal structures of *Galbella* that can be attributed to Schizopoid type most resemble the primitive states in some representatives of Agriloid complex, among them *Synechocera* DEYROLLE, 1864, *Polyonychus* CHEVROLAT, 1837, *Endelus* DEYROLLE, 1864, and *Germarica* BLACKBURN, 1887. On the other hand, the apical organs of *Galbella* and the particularly long sensillae uniformly dispersed over the antennomere surface in *G. felix* (Figs 217–219) are similar to those in some Acmaeoderioid taxa, primarily to Mastogeniini (Figs 10, 11). Unfortunately, such a similarity, resulting from symplesiomorphy, does not clarify possible relationships of *Galbella* to other higher taxa of Buprestidae. Some conclusions on the taxonomic position of Galbellinae, based on larval characters, will be drawn in the next chapter.

V. Taxonomical implications

See chapter II. (A list of examined buprestid higher taxa arranged with respect to antennal structures) for taxonomic changes suggested in this section.

Schizopoid complex (Schizopodinae)

General antennal structure shows that this complex is quite isolated and primitive while its complete compatibility with that of other Buprestidae does not support the elevation of Schizopodinae to family rank (NELSON & BELLAMY 1991), at least based on the current concept of subfamilies in Buprestidae. Distinctions in the evolutionary trends of anntenal structures confirm that Schizopodinae consist of two lineages deserving of tribal rank, i.e. Schizopodini (most primitive) and Dystaxiini (more advanced). The scale of the differences between *Glyptoscelimorpha* and *Dystaxia* that reveal the different pathways of sensory organ genesis corresponds to subtribal level in other buprestid taxa.

Julodoid complex (Julodinae)

Though the sensory organs of Julodinae are unique to this complex, I failed to recognize separate lineages or groups based on antennal structures because of their uniformity. Antennal characters fully contraindicate the transfer of Julodini to Buprestinae (KUROSAWA 1988); this is also confirmed by genital and larval (BÍLÝ 1983) structures.

Polycestoid complex (Polycestinae)

Although the general structure, genesis, and specialisation trends of antennal organs in Polycestinae are in many respects similar to those in Buprestoid complex, they demonstrate more primitive states producing their own morphoclines. Antennal, male genital, and larval (VOLKOVITSH & HAWKESWOOD 1999) characters of Polycestinae suggest this complex is a distinct subfamily, in contradiction to HOŁYNSKI (1988) who integrates Polycestinae with Buprestinae.

The antennal structure of Mastogeniini is closely similar to that of Acmaeoderini and Paratrachyini. Mastogeniini also share with Acmaeoderini genital characters, posterior pronotal margin straight, etc., However their closest affinity needs further investigation. I agree with an earlier assumption of HOŁYNSKI (1988) about the tribal rank of this taxon but disagree with his later downgrading of Mastogeniini to subtribe within Thrincopygini (HOŁYNSKI 1993) which is in conflict with above-mentioned characters.

Quite similar antennal structures in Acmaeoderini and *Odettea* support their close relationship. The main evolutionary trend in Acmaeoderini is development and perfection of a peculiar acmaeoderoid type of flight, which is characterised by a functional two-winged state and is accompanied by a profound modification of the entire pterothorax (VOLKOVITSH 1979, VOLKOVITSH & BELLAMY 1992), including: straightening of pronotal posterior margin and development of cremaliera for elytral base fixing; locking of elytral suture; reduction of scutellum, mesonotum, and subhumeral lobe of elytra; and modification of wing venation together with radial cell reduction throughout. Different stages of the transformation of these characters can be observed in all the groups of Acmaeoderini, from primitive states in *Odettea* and *Acmaeoderoides*, through intermediate ones in *Nothomorphina*, *Cochinchinula* VOLKOVITSH, 1984, and *Brachmaeodera* to the most advanced states in *Acmaeodera* (*Ptychomus*), *Xantheremia*, and *Acmaeoderella*. The similarity of Acmaeoderini to other Polycestoid taxa in antennal, genital, and larval (VOLKOVITSH & HAWKESWOOD 1999) structures as well as the markedly adaptive properties of many characters that are traditionally in use for delimitation of this group leave no grounds to regard Acmaeoderini as a separate subfamily.

Odettea was originally placed in Anthaxiini (BAUDON 1966, BELLAMY 1985). Later TOYAMA (1994) transferred it to Nothomorphini on the basis of wing venation, and finally

Bellamy & Bílý (1997) placed this genus in Polycestina using cladistic analysis. As well as the antennal organs *Odettea* shares with Acmaeoderini similar frontal and clypeal structure, head and pronotal sculpture, entirely serrate epipleura, elytral pattern and sculpture, scale pubescence, appendiculate tarsal claws, and adult anthophily; the only characters shared with Polycestini are curved pronotal posterior margin, wing venation, and dimorphism in elytral pattern, which are presumably symplesiomorphic. On the basis of the characters mentioned, I place *Odettea* in Acmaeoderini as a separate, extremely primitive subtribe Odetteina nov.

The antennal states of *Nothomorphoides*, *Nothomorpha*, and *Acmaeoderoides* integrated by Holm (1986) into Nothomorphini are identical to those in *Acmaeodera* and related genera. These taxa share the same symplesiomorphies that are represented in *Odettea* and *Ptosima*, such as visible scutellum, free elytral suture, and well developed subhumeral lobe (the latter occurs in *Cochinchinula* as well). *Acmaeoderoides* differs in having radial cell on hind wings present and some further characters that contributed to its placing in Ptosimina (Hołynski 1993, Bellamy & Westcott 1996). I presume that African *Nothomorphoides* and *Nothomorpha* on the one hand, and American *Acmaeoderoides* and recently described *Paracmaeoderoides* (Bellamy & Westcott 1996) on the other, form two subtribes within Acmaeoderini or two independent generic groups within Acmaeoderina. Both antennal and genital structures deny incorporation of Australian *Xyroscelis* in Nothomorphina, as well as the placement of the latter subtribe in Thrincopygini (Hołynski 1993).

With respect to antennal structure, Ptosimini have been traditionally contrasted with Acmaeoderini, being brought together with Polycestini (Bellamy 1985) or placed in Thrincopygini (Hołynski 1993). In fact, the antennal organs of *Ptosima* are identical to those in Acmaeoderini. Moreover they share posterior pronotal margin straight, bearing the cremaliera, appendiculate tarsal claws, and similar genital and larval (Bílý 1972, Volkovitsh & Hawkeswood 1999) structures. Lack of pterothorax modifications and adult anthophily, as well as some larval distinctions, suggest that Ptosimini should be placed next to Acmaeoderini. The incorporation of Nearctic *Acmaeoderoides* in Ptosimini (Hołynski 1993; Bellamy & Westcott 1996), based on symplesiomorphy, seems quite questionable.

A high degree of specialisation in Paratrachyini is apparent not only through their antennal organs, but also in other adult and particularly larval (Bílý 1989; Volkovitsh & Hawkeswood 1999) characters reflecting adaptations to the miner habit, at least in *Paratrachys*, attesting to appropriacy of tribal rank for this taxon. Although some characters of *Sponsor* support its possible relations to Ptosimina, the antennal structure is the same as that in *Paratrachys*. Based on cladistic analysis Bellamy & Williams (1995) held Paratrachyina to be monotypic and *Sponsor* to be a member of Ptosimina, but both the procedural correctness of the character state evaluation and the results of its analysis cast some doubts upon their conclusions.

It is traditionally believed that monotypic tribe Thrincopygini is a primitive taxon although its antennal and larval (Bílý 1986, Volkovitsh & Hawkeswood 1999) states as well as its biology (Nelson 1980) are rather specialised. In spite of numerous autapomorphies, the elevation of Thrincopygini to a separate subfamily is unwarranted. HOŁYNSKI (1993) proclaimed the affinity of Thrincopygini to Polyctesini, downgrading the latter to a subtribe of the former but his understanding of the Thrincopygini contains an artificial grouping, particularly in that it includes Agaeocerina.

Antennal (especially in the structure of lateral fields) and larval characters (VOLKOVITSH & HAWKESWOOD 1999) are in conflict with the placement of *Chrysophana* in Polyctesini (BELLAMY 1985). Apparently *Chrysophana* along with *Beerellus* (NELSON 1982) form their own tribe of Polyctesioid lineage or subtribe of Polyctesini. HOŁYNSKI (1993) transferred these genera from Polyctesina to Bubastina (Buprestinae), but based on the above-mentioned characters their belonging to Polycestoid complex is beyond question. The differences in sensory organs between *Polyctesis* and *Schoutendeiastes* support the generic rank of the latter (HOŁYNSKI 1993).

Primitive sensory formations in *Bulis* support its elevation to at least subtribe level (BELLAMY 1995), although in my opinion it should be of the same tribal rank as Polycestini. *Bulis* differs from other Polycestioid taxa in having external basiconic sensillae dispersed over the surface, which is at variance with its placement in Acherusini (COBOS 1980) or Astraeusini (HOŁYNSKI 1993). The affinity of Bulina to Prospherini established on the base of cladistic analysis (BELLAMY 1995) is questionable, since the autapomorphy shared by both taxa (hypomera with scrobes to receive fore- and mid-legs) frequently appears in unrelated taxa.

The morphocline of sensory organ states attests to Polycestini as a monophyletic taxon and even enables us to separate generic groups that are not identical to those that HOLM (1982) established on the basis of wing venation. The placement of Polycestina in Buprestini (HOŁYNSKI 1993) or its integration with Thrincopygini (BELLAMY & BÍLÝ 1997) are in total conflict with many adult and larval (BÍLÝ 1989, VOLKOVITSH & HAWKESWOOD 1999) characters showing that Polycestini and Thrincopygini belong to different phyletic lineages.

The marked similarity in sensory organs and male genital structure (especially in the shape of basal lobe of tegmen) (COBOS 1980) support the close affinities of Tyndarini, Tylauchenini, Acherusini, and Pseudacherusini and conform with the incorporation of the three former taxa in Tyndarini as subtribes (HOŁYNSKI 1993) (Pseudacherusini was placed in group Incertae sedis) but their integration with Prospherina and Astraeina does not agree with my results. The *Tyndaris* larva closely resembles those of Polycestini, bearing some minor distinctions (VOLKOVITSH & HAWKESWOOD 1999). Concerning the position and status of these taxa, two possibilities can be considered: 1) the preservation of Tyndarini as a distinct American tribe and downgrading Tylauchenini, Acherusini, and Pseudacherusini to subtribe level, or 2) the integration of Tyndarina and Polycestina in Polycestini wherein the other Cobos tribes should be downgraded to generic groups.

The relationships and taxonomic positions of Australian Xyroscelini, Astraeusini and Prospherini are still not quite clear and merit further investigation. It is possible that they belong to an exclusive Australian lineage or lineages, yet their affinities to non-Australian taxa must not be ruled out. The sensory organs of *Xyroscelis* bear a certain similarity to those in Acmaeoderioid and Polyctesioid taxa. Based on habitual similarities, HOŁYNSKI (1993) placed *Xyroscelis* in Nothomorphina (Thrincopygini). Using cladistic analysis BELLAMY (1997) suggested that it is closest to *Ptosima*. However, *Xyroscelis* differs from other Polycestoid taxa in having a penis of an unique type, which confirms its separate position. Antennal and larval (VOLKOVITSH & HAWKESWOOD 1999) characters of *Prospheres* somewhat resembling those in Buprestoid taxa contraindicate its placement in Polyctesini (LEVEY 1978b) or Tyndarini (HOŁYNSKI 1993), however sensory organs confirm a close relationship of *Prospheres* to *Euleptodema* (LEVEY 1978b, HOŁYNSKI 1993). With the exception of the presence of distinct lateral depressions, the sensory organs of *Astraeus* are similar to those in Polyctesini (*Polyctesis*) which may or may not result from parallel evolution. The placement of Astraeusina integrated with *Bulis* in Tyndarini does not agree with our results.

Chalcophoroid complex (Chalcophorinae)

Although the integration of Chalcophorinae with Buprestinae is a currently prominent proposal (TOYAMA 1987, HOŁYNSKI 1988, 1993), the characteristic Chalcophoroid pathway of sensory organ genesis supports the separation of Chalcophoroid complex as a distinct subfamily which consists of well distinguished subordinate taxa.

The antennal organs of Vadonaxiini confirm that this monotypic and presumably relict taxon, with obscure affinities, is rather isolated.

The taxonomic position and relationships of *Paratassa* based on the analysis of adult and larval characters was recently discussed by BÍLÝ & VOLKOVITSH (1996). By virtue of the fact that sensory organ states and other features contraindicate the attribution of *Paratassa* to Bubastini (Buprestinae), the new tribe Paratassini of Chalcophorinae was established, contrary to general opinion.

Nanularia and *Ampheremus,* which are usually integrated with *Hippomelas* (BARR 1970, BELLAMY 1987), or considered to be closest to it, were recently placed in subtribe Hippomelanina of Buprestini (HOŁYNSKI 1993, NELSON & BELLAMY 1996). However, their sensory formations belong to different morphoclines, corresponding to two phyletic lineages. *Nanularia* and *Ampheremus* demonstrate one of the most primitive states of the Chalcophorioid morphocline whereas *Hippomelas* shows the primitive state of Psilopterioid morphocline. That these genera belong to different lineages is also confirmed by larval structure (BÍLÝ & VOLKOVITSH 1996, NELSON & BELLAMY 1996), wing venation and other characters which suggest that *Nanularia* and *Ampheremus* form their own separate group near the base of Chalcophorioid lineage. Antennal structure also supports the generic status of *Ampheremus*.

Afrotropical *Chalcoplia* used to be placed in Chalcophorini (BELLAMY 1985, HOŁYNSKI 1993) but recently HOŁYNSKI (1997) transferred it to Chrysochroina together with Australian *Chalcophorotaenia* OBENBERGER, 1928 and *Chalcomroczkowskia* HOŁYNSKI 1997 as its subgenera. Although I did not study the two latter taxa, the antennae of the genera that HOŁYNSKI (1997) attributes to Chrysochroina and places next to *Chalcoplia* (*Metataenia*, *Papuodema*, *Cyphogastrella*, *Cyphogastra*, *Callistroma*, *Paracupta*, etc.) differ greatly from those in the examined *Chalcoplia auripilis*. The poorly known African *Descarpentriesiola* (= *Castalina* THÉRY) was transferred from Polycestinae to Chalcophorini by COBOS (1978) who indicated its possible affinity to *Chalcoplia*. *Embrikillium* was set up as a distinct genus but was later downgraded to a subgenus of *Oedisterna* (Psilopterini) (BELLAMY 1985); then BELLAMY (1988) resurrected its generic rank and using antennal structure, among other characters, transferred this genus to Chalcophorini and noted its similarity to some species of *Chalcoplia*. In spite of a certain similarity of the sensory organs, its placement in Pseudoperotina (HOŁYNSKI 1993) is disallowed by the fact that in *Embrikillium* the zones of U2 sensillae are absent. Similarities in antennae (enlarged from strongly elongated 3rd antennomere which is much longer than 2nd and 4th, sensory organs appearing from 4th antennomere), the shape of body and pronotum, the strongly flattened sides of pronotum, the shape of scutellum, and long wavy hairs prove the affinity of *Embrikillium* to *Descarpentriesiola*. I presume that the sensory organ states of *Chalcoplia*, *Embrikillium*, and *Descarpentriesiola* produce the peculiar morphocline and these genera form a separate Afrotropical group of Chalcophorinae. The possibility must not be ruled out that African *Fahraeusia*, in spite of sharp distinctions in sensory organ structure, may be also associated with this group, since dense, long U2 sensillae on top of the last antennomere and the body shape are quite similar to *Embrikillium*.

The problem of delimitation and taxonomical status of Chrysochroini and Chalcophorini, which are undoubtedly closely related groups, has been repeatedly discussed in recent years (LEVEY 1978a, TOYAMA 1987, KUROSAWA 1990, HOŁYNSKI 1993, 1997). LEVEY (1978a) made an attempt to find reliable diagnostic characters to separate these taxa but as HOŁYNSKI (1993) correctly indicated, none of the suggested characters provides a way to draw a distinct line between them. TOYAMA (1987) integrated Chalcophorinae with Buprestinae and HOŁYNSKI (1993, 1997), who has supported this opinion, regarded both taxa as either distinct tribes of Buprestinae or subtribes of Buprestini. HOŁYNSKI (1993) maintained that the traditional delimitation of Chrysochroini and Chalcophorini based on the presence or absence of visible scutellum and antennal structure is artificial but he failed to suggest any alternative characters.

The examination of antennal structure in these groups has shown that: 1) the presence or absence of sensory fossae in Chrysochroini and Chalcophorini, which appear occasionally as a result of the confluence of pores or small pits and have no fixed position, is not a reliable character with which to separate not only the tribes but even generic groups (LEVEY 1978a); 2) Chrysochroini and Chalcophorini are recognised not by antennal organ states but by directions of antennomere shape variability which lead towards flabellate antennae in *Agelia* (Chrysochroini) or long, serrate, highly specialised antennae in *Chalcophora* (Chalcophorini); 3) the borderline between Chrysochroini and Chalcophorini apparently draws near *Cyphogastra* and *Chrysodema* generic groups (HOŁYNSKI 1993). Failure to find more reliable diagnostic characters has led me to accept Chrysochroina and Chalcophorina as subtribes of the same tribe, which is supported by the similarity in larval structures (for example, the presence of big asperities covering the prothoracic plates). The priority and authorship of the tribal name need further clarification. For the moment, I retain the traditional name Chalcophorini. In doing so I regard Chrysochroina as including only *Callopistus* and *Chrysochroa* generic groups.

I do not support the separation of the genera with antennae widened from 3rd antennomere into the separate subtribe Callopistina, in contrast to Chrysochroina in which the antennae widen from 4th onwards (KUROSAWA 1990), since in *Epidelus*, *Philocteanus*, and *Cyalithus* the normal 3rd antennomere bears a rudimentary sensory field internally. Otherwise antennal structures of Callopistina are identical to those in other chrysochroine genera, except the modified antennae of *Philocteanus* and *Micropistus*. It is also hard to agree with the generic composition and status of Chalcophorellini (TOYAMA 1986) and Iridotaenini (TOYAMA 1987) which were erected exclusively on the basis of wing venation. The antennal state in *Iridotaenia* fits better in chrysochroine morphocline, whereas that in *Metataenia* corresponds to the chalcophorine one. Further, HOŁYNSKI (1993) correctly indicates that *Iridotaenia* is assumed to be a heterogenous taxon. The placement of undoubtedly closely related *Stigmatophorella* and *Rosiella*, usually regarded as subgenera of *Chalcophorella*, in different tribes is not supported by similarity of antennal structures. *Nipponobuprestis* and *Texania,* which are sometimes placed next to *Chalcophorella* (OBENBERGER 1942), are actually related to *Chrysodema* group, which with *Texania* apparently forms a separate monotypic Nearctic group. The antennal structure of Oriental *Rhabdolona*, which was recently transferred on the basis of wing venation from Dicercini to Buprestini (TOYAMA 1987) shows that it should be placed in Chalcophorini. I did not study characters other than antennae, but on the basis of antennomere shape and very large depressions of confluent pores and pits, *Rhabdolona* is similar to *Sapaia* and Genus 1 which possibly form a separate generic group. On the basis of antennal structures, *Cyphogastra* (*Cyphogastra*, *Cyphogastrella*, *Callistroma*, *Paracupta*, *Papuodema*), *Chrysodema* (*Chrysodema*, *Thymedes*, *Tamamushia*, *Nipponobuprestis*, *Metataenia*, *Gelaeus*), *Chalcophoropsis* (*Chalcophoropsis*), *Scaptelytra* (*Scaptelytra*), and *Pseudotaenia* (*Pseudotaenia*) generic groups may be also recognised. The antennae of *Lampropepla* which, together with *Chalcophoropsis* and *Afrophorella*, was separated by HOŁYNSKI (1993) into Lampropeplina, were not studied but the antennal state of *Chalcophoropsis* proved much closer to those in *Chrysodema* group, and that of *Afrophorella* to *Pseudotaenia*, *Chalcotaenia*, *Austrophorella*, and *Chalcophora*. The last mentioned, demonstrating the most specialised antennal structures among Chalcophorina, can be separated into its own group next to *Pseudotaenia* and *Chalcotaenia*. Finally, *Chlorophorella*, in which antennal structure differs greatly from that of other genera, should be separated into a distinct group or even subtribe.

The monotypic tribe Evidini erected by TOYAMA (1987) on the basis of antennal structure and wing venation was placed next to Iridotaenini. The close relationship of *Evides* to Chrysochroina – Chalcophorina is beyond question and it is quite possible that Evidini should be regarded as the most specialised subtribe of Chalcophorini.

Neotropical genera *Euchroma*, *Hypoprasis*, *Baudonisia*, *Halecia*, *Eupodalecia*, *Pelecopselaphus*, *Chrysesthes*, *Euplectalecia*, and *Saundersina* are usually attributed to Chalcophorini (COBOS 1957, BELLAMY 1985, 1986b) whereas *Cinyra* along with

Australian *Euleptodema* (currently placed in Prospherini) was placed in the separate tribe Cinyrini (COBOS 1979b) (= Paraleptodemini COBOS, 1975a). TOYAMA (1987) transferred *Pelecopselaphus* to Agaeocerini (Buprestini), mainly on the basis of wing venation. HOŁYNSKI (1993) placed it in the new subtribe Pristipterina of Buprestini along with *Pristiptera* (= *Halecia*), *Chrysesthes*, and *Saundersina*. He also established the new subtribes Euchromatina, Hypoprasina, and Euplectaleciina and synonymised Cinyrini with Phrixiina. BELLAMY & WESTCOTT (1995), being doubtful of the validity of the name *Pristiptera*, added *Eupodalecia* and *Pseudalecia* to *Halecia* generic group (= Pristipterina). Since the sensory organ states of all the above-mentioned taxa produce a peculiar morphocline which only to some extent coincides with those in Chalcophorini and Psilopterini I suggest that these genera be placed in the separate Neotropical tribe Hypoprasini HOŁYNSKI, 1993, **stat.nov.** comprising several subtribes and generic groups as follows: Euchromatina (*Euchroma*), Hypoprasina (*Hypoprasis*, *Baudonisia*, *Cordillerita*), Pristipterina (*Halecia*, *Eupodalecia*), Euplectaleciina (*Euplectalecia*), Cinyrina COBOS **stat.nov.** (*Cinyra*), *Pelecopselaphus* group (*Pelecopselaphus*, *Chrysesthes*), and *Saundersina* group (*Saundersina*); perhaps the final two groups may also be treated as separate subtribes. Antennal structures of *Cordillerita* prove that this genus should be transferred from Dicercini to Hypoprasina.

Poecilonota and *Palmar* are either integrated with *Dicerca* in Psilopterini (= Capnodini JACOBSON, 1911) (RICHTER 1952, HOŁYNSKI 1993) or placed in Dicercini (Buprestinae) (BELLAMY 1985). RICHTER (1952) indicated that *Poecilonota* and *Palmar* (as *Lampra*) differ from *Dicerca* in wing venation, large scutellum, and larval pronotal groove configuration. ALEXEEV & BEBKA (1970) established that *Poecilonota* and *Plamar* (as *Lampra*) have 7 external tergites in contrast to 8 in *Dicerca* and erected the separate tribe Poecilonotini within Ancylocheirini JACOBSON, 1911 (= Buprestinae); *Dicerca* was placed in Capnodini of Buprestinae (= Chalcophorinae). Although the reduction of external tergite number resulted from the 8th tergite's incorporation into perigenital structures that apparently appear in parallel in different, non-related taxa, the distinctive morphocline of antennal states, together with other characters, supports the separation of Poecilonotini as a distinct tribe. In spite of an external resemblance in antennal structure, the complete lack of U2 sensilla zones in Poecilonotini, in contrast to their being well developed in *Dicercomorpha* and *Haplotrinchus*, indicates that the first belongs to Chalcophorioid and the latter two to Psilopterioid lineage. An obvious resemblance in antennal organs between Poecilonotini and *Nesotrinchus* together with serrate epipleura and similar frontal sculpture support their possible relationship and argue against the placement of *Nesotrinchus* (as Melobasina, part.) in Haplotrinchina (HOŁYNSKI 1993).

Although the separation of Sphenopterini is beyond any doubt, their taxonomical position, relationships, and status require further clarification. Many authors regard *Sphenoptera* and related genera as a distinct tribe or even subfamily, placed between Chalcophorinae and Buprestinae (COBOS 1980, 1986a, BELLAMY 1985). HOŁYNSKI (1988, 1993), without adducing any proofs, placed subtribe Sphenopterina in Anthaxiini. Antennal organs and larval characters are in complete disagreement with this idea, indeed giving an indication that Sphenopterini belong to Psilopterioid lineage of Chalcophoroid complex. If this assumption were not supported by other characters, Sphenopterini should be placed at the base of Chalcophoroid phyletic stock next to Paratassini and *Nanularia-Ampheremus* group.

The composition and status of Psilopterini and its subordinate taxa are a matter of perennial debate. HOŁYNSKI (1993) correctly points out the unreliability and variability of the diagnostic characters in great part based on antennal structures that are traditionally in use for delimitation of Chalcophorini and Psilopterini. Indeed, this research has shown that sensory organ states sometimes coincide almost completely in both phyletic lineages but the presence of fields of U2 sensillae is inherent in psilopterine taxa. Psilopterini are also clearly different from Chalcophorini in larval characters (lack of sclerotised asperities on prothoracic plates). As mentioned above, the different groups of Psilopterini might have evolved independently in the Old and New Worlds, with Madagascar and possibly South Africa hosting separate generic groups or subtribes.

Hippomelas together with *Nanularia* and *Ampheremus* were attributed to Chalcophorini (BELLAMY 1985), later HOŁYNSKI (1993) erected subtribe Hippomelanina of Buprestini which was supported by NELSON & BELLAMY (1996) although they did not accept the downgrading of *Nanularia* and *Ampheremus* to subgenera of *Hippomelas*. As discussed earlier, these genera form their own group within Chalcophorioid lineage. Antennal organs of *Hippomelas* and related genera (NELSON & BELLAMY 1996) indicate that they certainly belong to Psilopterioid lineage, which is also confirmed by larval characters. Differences in proportions of proximal antennomeres and the shape of the apical one suggest that Hippomelanina may be treated as a distinct subtribe of Psilopterini.

The relative primitiveness of sensory organs, together with other characters, supports the separation of Pseudoperotina (TOYAMA 1987, HOŁYNSKI 1993) as a distinct subtribe. Antennal structures of *Asidoptera* and *Pseudoperotis* are completely identical.

According to genotype designation by KUROSAWA (1993), *Psiloptera* is an exclusively American genus. Some American species usually attributed to Old World *Lampetis* (COBOS 1954, NELSON 1982) apparently form a separate subgenus or subgenera of *Psiloptera*. COBOS (1972) and HOŁYNSKI (1993) regard *Achardella* as a subgenus of *Ectinogonia* although the poor oligomerization of antennal organs, in contrast to *Ectinogonia*, supports its generic rank. *Chalcopoecila* was placed by KERREMANS (1903) in Dicercini; COBOS (1954) pointed to its similarity to Psilopterini but latter he placed this genus in Chalcophorini (COBOS 1957); TOYAMA (1987) transferred *Chalcopoecila* from Dicercini to Buprestini on the basis of wing venation; finally HOŁYNSKI (1993) placed it in the Afrotropical Pseudoperotina. Based on the presence of U2 sensillae and short 3rd antennomere this genus belongs to Psilopterini; its antennal organs, which closely resemble those in *Ectinogonia*, support the placement of *Chalcopoecila* in *Psiloptera* generic group and attest to it as a highly specialised genus.

Marked differences in antennal structures between the two species of *Polybothris* studied show that this huge genus is heterogenous, a fact also confirmed by external characters. The isolation and abundance of *Polybothris* species support an assumption that Madagascar is one of the largest centres of speciation. From my viewpoint, Madagascan *Polybothris-Icarina* form a separate generic group of the Psilopterina or even a separate subtribe of Psilopterini.

On the basis of the structure of lateral organs, two minor groups can be recognised within the Old World *Lampetis* generic group. Those are *Capnodis* and *Cyphosoma*, having lateral fields of mainly single pores and small pits, and *Lampetis*, *Perotis* and *Oedisterna* bearing well developed lateral fossae.

This study has shown that Dicercini (BELLAMY 1985) is an entirely polyphyletic group comprising both Chalcophoroid and Buprestoid taxa, and confirmed that *Dicerca* belongs to Psilopterini as RICHTER (1952) and HOŁYNSKI (1993) stated. It is presumed that *Dicerca* together with *Latipalpis* forms a separate group of Psilopterina characterised by strong oligomerization of sensory organs and transformation of antennomere shape. It is quite possible that *Dicercomorpha* is closely related to this group but this assumption requires further investigation.

In spite of the fact that the sensory organs of *Cardiaspis* and *Haplotrinchus* greatly resemble those in *Dicercomorpha*, I retain them within Haplotrinchina in accordance with the opinion of HOŁYNSKI (1993) until new data becomes available. As noted above, *Nesotrinchus* should be removed from this subtribe.

Buprestoid complex (Buprestinae)

It is hard to judge the real structure of *Mendizabalia* sensory organs while the female remains unexplored; however, the marked differences in the male antennae between *Mendizabalia* and *Philandia* cast some doubts upon the monophyly of Mendizabaliini. This seems contradictory to the placement of *Philandia* in Mendizabaliini based in part on external similarity in antennal characters (BELLAMY & MOORE 1991). This assumption is also supported by the differences in pronotal structure and wing venation. Nevertheless, available data does not allow the unequivocal determination of the position and affinities of these genera, apart from the fact that they belong to Buprestoid rather than Chalcophoroid complex as was maintained by COBOS (1968) and BELLAMY (1985).

The taxonomic position and relationships of *Maoraxia* (described as *Maoriella*, nom. praeocc.) raise heated debates (see BELLAMY & WILLIAMS 1985). Originally it was placed in Mastogeniini, which has been regarded as closest to Agrilinae (OBENBERGER 1924, 1937), although many authors placed *Maoraxia* (sometimes as *Neocuris*, part.) in Anthaxiae CARTER & THÉRY. Recently HOŁYNSKI (1984) separated *Maoraxia* from Mastogeniini in the monotypic tribe Maoraxiini. BELLAMY & WILLIAMS (1985) treated *Maoraxia* as a member of Anthaxiae giving it proximity to *Neocuris* and *Pseudanilara*. Later BELLAMY (1986a) synonymised Maoraxiini with Anthaxiini. Finally, HOŁYNSKI (1988) resurrected Maoraxiina as a subtribe of Anthaxiini, which is currently accepted (BELLAMY 1990, 1991c, HOŁYNSKI 1993). The examination of the antennal structures of *Maoraxia* has shown that there is a certain similarity with those in Mastogeniini, although male genital (BELLAMY & WILLIAMS 1985, BELLAMY 1990, 1991c) and larval (DUMBLETON 1932, as *Neocuris*) characters in *Maoraxia* indicate that it belongs to Buprestoid complex. However, the sharp differences in antennal structures along with some adult (fronto-clypeal carinae, wing venation) and larval (characteristic 2nd segment of maxillary palpi, densely microspinulated antennae, labrum and prementum, prothoracic plates and abdominal segments bearing asperities) characters contraindicate the placement of *Maoraxia* in Anthaxiini. On the other hand, such larval features as inverted V-shaped pronotal grooves and the presence of both dorsal and ventral ambulatory pads on the sides of metathorax being evidence of its possible affinities to Anthaxiina whereas prosternal groove bifurcated at posterior 2/3 indicating *Melobasis*, suggest that *Maoraxia* might be a sister group for Anthaxioid taxa. I cannot comment on the relations of *Maoraxia* to *Australorhipis* BELLAMY, 1990 but according to wing venation (BELLAMY 1986a, 1990) this is quite possible. It is supposed that Maoraxiini is a distinct relict tribe that should be placed at the base of Buprestoid phyletic stock; the above-mentioned characters do not allow its certain attribution to either Buprestioid or Anthaxioid lineage.

In spite of the fact that the sensory organs of *Julodimorpha* consist of confluent fragments, a characteristic of Chalcophoroid taxa, their general structure suggests it is more likely to belong to Buprestoid complex. BELLAMY (1986a) placed *Julodimorpha* in Chalcophorinae and indicated its isolated position. I presume that Julodimorphini, on the basis of similarity in sensory organs to Epistomentini and Buprestini, belong to or are closely related to Buprestioid lineage, whereas the above-mentioned difference along with other characters attest to this tribe's comparative isolation. This accords with HOŁYNSKI (1993) who preserved its tribal rank and uncertain position. The antennal structure of Julodimorphini has nothing in common with that of Julodinae.

Nascio and *Nascioides* have usually been placed in Buprestini (BELLAMY 1985). KUROSAWA (1988) brought *Nascio* together with Nearctic *Trachykele*, a move that is not supported by antennal structures. HOŁYNSKI (1988, 1993) integrated *Nascio* and *Nascioides* with Neotropical *Pterobothris* in subtribe Nascionina of Anthaxiini. Based on both antennal and larval characters (see below) *Pterobothris* differs greatly from these genera. There are also significant differences between antennal organs of *Nascio* and *Nascioides* that, along with other characters (WILLIAMS 1987), cast some doubt upon their close affinity. Although the sensory organs of *Nascioides* bear a certain similarity to those in *Coomaniella* and *Kisanthobia*, its placement in Anthaxioid lineage is completely unsupported by larval characters (DUMBLETON 1932, VOLKOVITSH, unpublished data) which resemble *Pygicera* and *Diadoxus*. The low specialisation level of both antennal and larval characters suggests that Nascionina should be placed at the base of Buprestioid lineage.

Pygicera has sensory organs somewhat resembling those in *Bulis* from Polycestinae, but larval characters are different from those in other Buprestioid taxa (VOLKOVITSH, unpublished data). On this basis, *Pygicera* cannot be included (as suggested by HOŁYNSKI 1993) in *Epistomentis* as subgenus and it should apparently be separated into its own group.

Epistomentini were established as a tribe of Chalcophorinae (LEVEY 1978a); however, the similarity of its sensory organs to those in Australian *Julodimorpha*, *Neobuprestis*, and *Balthasarella*, as well as to *Buprestis* and related genera, proves their placement in Buprestioid lineage. Accordingly, Epistomentini should be transferred to Buprestinae, a move supported by larval characters of *Diadoxus* (HAWKESWOOD 1985, VOLKOVITSH & HAWKESWOOD 1993, 1994). Differences between *Cyria* and *Xenocyria* in the shape of distal antennomeres and sensory organs confirm the generic rank of the latter.

Neobuprestis was originally placed in Dicercites (Dicercini) (KERREMANS 1903, BELLAMY 1985) whereas *Balthasarella* has been regarded as a member of Polycestinae (OBENBERGER 1958). Subsequently *Neobuprestis* was transferred to Buprestini (BELLAMY 1986a) and then placed in *Buprestis* generic group next to *Cypriacis* CASEY, 1909 (KUROSAWA 1988); COBOS (1974) assumed that *Balthasarella* also belongs to Buprestini. BELLAMY (1991a) pointed out the possible affinity of newly described African *Zulubuprestis* to *Neobuprestis* and *Balthasarella* and after examination of the wing venation of the latter placed all these genera in *Buprestis* group (BELLAMY 1994). HOŁYNSKI (1993) treated *Neobuprestis* as a subgenus of *Microcastalia* of Polycestina (Buprestini), which is not confirmed by my results. Although the antennal structures of *Neobuprestis* and *Balthasarella* partly support their affinities to Buprestini, they demonstrate great similarity to those in Julodimorphini and Epistomentini. Sharp antennal dimorphism, at least in *Neobuprestis*, and female sensory organs that much resemble those in Prospherini, more primitive when compared with *Buprestis* group, suggest that *Neobuprestis* and *Balthasarella* constitute a separate Australian group.

KUROSAWA (1988) brought *Trachykele* together with *Nascio* primarily on the basis of similar pronotal shape. HOŁYNSKI (1988, 1993) erected monotypic subtribe Trachykelina of Anthaxiini. However, *Trachykele*, based on similar antennal and larval characters, is much closer to *Buprestis* and related genera than to both *Nascio* and Anthaxiini, so Trachykelina should be transferred to Buprestini.

The composition of subtribe Buprestina after the removal of *Neobuprestis* and *Balthasarella* (and possibly *Zulubuprestis*) is almost beyond question but the status of remaining taxa is still controversial. In spite of similar antennal structure, the differences in the level of differentiation of the apical organs and lateral fields in studied species of *Buprestis*, *Eurythyrea*, and *Yamina*, along with larval characters, attest to these as a good genera, contrary to HOŁYNSKI (1993) who regards them as subgenera of *Buprestis*.

Lamprocheila was attributed to Chalcophorini (BELLAMY 1985) but recently HOŁYNSKI (1993) erected monotypic subtribe Lamprocheilina of Buprestini and placed it between Hippomelanina and Psilopterina, indicating its possible relations to *Dicerca* and *Palmar*. With respect to antennal structure, *Lamprocheila* belongs to Buprestioid lineage but its affinities are not quite clear. Having no other arguments I retain Lamprocheilina as a subtribe of Buprestini.

Tribe Agaeocerini was originally established for Nearctic *Agaeocera* within Buprestinae next to Dicercini (NELSON 1982), while the possible affinities of *Agaeocera* to *Mixochlorus* were also proclaimed. TOYAMA (1987) transferred *Pelecopselapus* to this tribe but, as noted above, it belongs to Chalcophoroid complex. HOŁYNSKI (1993) downgraded Agaeocerini comprising *Agaeocera*, *Mixochlorus* and *Peronaemis* to a subtribe of Trincopygini (Polycestoid complex). BELLAMY & BÍLÝ (1997) suggested that it should be transferred to Buprestini and restored *Pelecopselaphus* along with *Chrysesthes* as its members. Sensory organ states of *Agaeocera* which completely fit the Buprestioid morphocline confirm its close affinity to *Mixochlorus* and *Peronaemis* but not to Polycestoid taxa or *Pelecopselaphus* and *Chrysesthes*. The similarity in antennal structure to *Buprestis* and related genera supports the transfer of Agaeocerina from Trincopygini to Buprestini.

Exagistus has been placed in Melanophilini (BELLAMY 1985). TOYAMA (1987) erected monotypic tribe Exagistini next to Psilopterini and Dicercini on the basis of wing venation. HOŁYNSKI (1993) transferred *Exagistus* to Phrixiina of Buprestini. In view of its antennal structure *Exagistus* has nothing in common with either Melanophilini, or Psilopterini and *Dicerca*, and differs significantly from Phrixiini, resembling *Peronaemis* of Agaeocerina. However, its direct affinity to the latter seems very questionable and I regard Exagistini as a separate tribe next to Buprestini until additional evidence becomes available.

Phrixia and *Paraphrixia* were previously placed in Polycestinae (OBENBERGER 1926). COBOS (1975a) erected the tribe Phrixiini and transferred it to Chalcophorinae. HOŁYNSKI (1993) downgraded Phrixiini to a subtribe of Buprestini and added to it such unrelated genera as *Aglaostola*, *Cinyra*, *Spectralia*, and *Exagistus*. Considering antennal structures, Phrixiina sensu HOŁYNSKI is an absolutely artificial taxon. The sensory organ states of *Phrixia* and *Paraphrixia* correspond to Buprestioid morphocline whereas the existing distinctions support the tribal rank of Phrixiini which, however, together with its position, invites further investigation.

Australian genus *Buprestina* was placed in Buprestini (BELLAMY 1985). Its characteristic feature is modified B4a sensillae which are also found in some American genera. Otherwise the sensory organ structure is similar to that in *Bubastes* and *Euryspilus* (Bubastini), the shape of elytral apices bearing some resemblance to that in the latter and *Microcastalia*. The above-mentioned characters are inadequate to clarify the position of *Buprestina*; it can be separated in its own group close to Bubastini on the basis of antennal characters.

Bubastini sensu OBENBERGER (1920) included several Australian genera, Afrotropical *Strandiola* OBENBERGER, 1920, and Palaearctic *Paratassa*. HOŁYNSKI (1988) downgraded Bubastini to a subtribe of Anthaxiini removing *Notobubastes* CARTER, 1924. Later HOŁYNSKI (1993) synonymised Thomassetiina and Philanthaxiina with Bubastina and also transferred *Chrysophana* there from Polycestoid complex, thus creating an absolutely artificial conglomeration of unrelated taxa. BÍLÝ & VOLKOVITSH (1996) separated *Paratassa* into monotypic tribe Paratassini and transferred it to Chalcophorinae. The comparison of antennal structures of Australian Bubastini has shown that it is made up of two generic groups, *Bubastes-Euryspilus* and *Neobubastes-Eububastes*. The sensory organ states of the former correspond to Buprestioid morphocline bearing a certain similarity to *Buprestina* and *Microcastalia*; those in the second group fit into Anthaxioid morphocline resembling the states in Curidina, Thomassetiini, and Philanthaxiina. The polyphyly of Bubastini is also supported by the configuration of the pronotal keels and elytral apices. I suggest that Bubastini, comprising *Bubastes* and *Euryspilus*, belongs to Buprestioid lineage, being closely related to *Buprestina*, *Microcastalia*, and Stigmoderini. *Neobubastes* and *Eububastes* form their own group within Anthaxioid lineage and should be removed from Bubastini. *Strandiola* was not studied.

The position and affinities of *Microcastalia* remain uncertain. Kerremans (1902) and Obenberger (1926) placed it in Polycestinae which was supported by Hołynski (1993) who has attributed *Microcastalia* (including subgenus *Neobuprestis*) to Polycestina of Buprestini. Cobos (1978) downgraded *Ditriaena* which was previously transferred from Dicercini to Buprestini (Cobos 1975b) and *Goryola* (Cobos 1978) to subgenera of *Microcastalia*. In antennal structure, which certainly belongs to Buprestoid type, *Microcastalia* has nothing in common with Polycestini but it also differs greatly from *Ditriaena* (*Goryola* was not studied) or *Neobuprestis*. Apical organs of *Microcastalia* are to some extent similar to those in genus *Buprestina* and Bubastini whereas their position, as well as the position and composition of lateral fields, resembles that in *Calodema* (Stigmoderini). Based on antennal characters *Microcastalia* should be separated in a separate Australian group next to *Buprestina*, Bubastini, and Stigmoderini.

The taxonomy and phylogeny of Stigmoderini based on detailed analysis of morphological and anatomical characters, including antennal structures, is described in an excellent revision by Gardner (1989). Unfortunately, the relationships of Stigmoderini were not discussed. Antennal characters indicate Stigmoderini are presumably related to *Buprestina*, Bubastini, and *Microcastalia*. Gardner (1989) treated *Hypostigmodera* as a subgenus of *Castiarina* but, on the basis of the antennal structures of both sexes, which differ greatly from those in other Stigmoderini, it deserves generic rank.

Aglaostola was placed in Dicercini (Buprestinae) (Bellamy 1985), then Hołynski (1993) transferred it to Phrixiina. Its antennal organs differ greatly from those in both *Dicerca* and Phrixiini. Although the modified B4a sensillae within apical fossae of *Aglaostola* resemble those in Australian genus *Buprestina* and American *Hilarotes*, *Spectralia*, *Ditriaena*, and *Pterobothris*, such a similarity may result from parallel evolution. Having no other evidence, I separate *Aglaostola* into its own group close to *Hilarotes*, *Spectralia*, and *Ditriaena*.

The position of *Hilarotes* in Chalcophorini (Bellamy 1985) was recently disputed by Hołynski (1993) who placed it in Psilopterina next to *Touzalinia* and *Dicercomorpha*, although noting that its tarsal structure is typical for Chalcophorini. *Spectralia* and *Ditriaena* (as a subgenus of *Microcastalia*) were placed in Dicercini (Bellamy 1985) although Cobos (1975b) had earlier transferred *Ditriaena* to Buprestini, retaining *Spectralia* (as *Cinyra* auct., nec Laporte & Gory) in Dicercini. Hołynski (1993) placed *Spectralia* in Phrixiina (Buprestini) making no mention of *Ditriaena*. The extensive lateral fields of external B4c and U2 sensillae suggest that these genera may belong to Psilopterioid lineage of Chalcophoroid complex whereas the presence of modified B4a sensillae in apical fossae points to their possible affiliation to Buprestoid complex. *Hilarotes* differs in habit from both *Spectralia* and *Ditriaena* whose affinity is almost beyond question. As soon as these relationships are supported with other than antennal characters, the above-mentioned genera should be elevated to a separate tribe of Buprestioid lineage close to *Aglaostola*, and, apparently, to *Buprestina* and *Pterobothris*. Yet, based on resemblance in lateral fields, their possible relations to Psilopterini must not be completely ruled out. The position and relationships of these genera needs further investigation along these lines.

Pterobothris is usually placed in Buprestini (BELLAMY 1985), HOŁYNSKI (1993), based apparently on its external similarity to *Nascio*, transferred *Pterobothris* to Nascionina of Anthaxiini. However, the sensory organs of *Pterobothris* have nothing in common either with those in *Nascio*, or with Anthaxiini, resembling (especially in well-defined lateral organs) some Agrilini LAPORTE, 1835 (*Callichitones* OBENBERGER, 1931, *Geralius* HAROLD, 1868). Modified B4a sensillae in apical fossae of *Pterobothris* look the same as those in *Buprestina*, *Aglaostola*, *Hilarotes*, *Spectralia*, and *Ditriaena*, although their close relationships should be proposed with caution. The similarity of *Pterobothris* to Agrilini is shown not only in antennal but also in larval characters (MOORE & CERDA 1986), particularly in typical agriline body terminated with strongly sclerotised supporting processes. However a preliminary study of the *Pterobothris* larva shows that both pronotal grooves and spiracles correspond to Buprestoid type. In spite of this, detailed additional study of adult and larval characters is urgently needed; it may be suggested that in all probability *Pterobothris* is a member of Buprestoid complex and if its affinities to *Aglaostola*-*Ditriaena* were to be confirmed, it should be treated as an isolated and highly specialised branch of Buprestioid lineage. In any case, *Pterobothris* must be elevated to the separate tribe Pterobothrini **trib.nov** on the basis of its antennal and larval characters**.**

As noted above, *Neobubastes* and *Eububastes*, with respect to their antennal and some other characters, differ significantly from *Euryspilus* and *Bubastes* and must be removed from Bubastini. Based on antennal similarity with Thomassetiini, *Aristosoma*, *Philanthaxia* and Curidina these genera should be transferred close to the above mentioned taxa belonging to Anthaxioid lineage. In spite of the resemblance in sensory organs to Curidina, their close relationship is in some doubt. The integration of *Neobubastes* and *Eububastes* with Thomassetiina and Philanthaxiina as HOŁYNSKI (1993) suggested needs further investigation. Until more data is provided I will treat these genera as a separate group.

Thomassetiina (as a tribe) was established for South African *Thomassetia*, *Augrabies*, *Jakovleviola*, and *Senegalisia* BELLAMY in BELLAMY et al. 1987 (BELLAMY et al. 1987). Three former genera were previously placed in Buprestini (BELLAMY 1985). HOŁYNSKI (1988) erected subtribe Philanthaxiina of Anthaxiini without considering its generic composition, then BELLAMY (1991b) synonymized it with Thomassetiina, and finally HOŁYNSKI (1993) integrated Thomassetiina and Philanthaxiina with Bubastina. Antennal structure supports the affinity of Thomassetiina to Australian *Neobubastes* and *Eububastes* (but not to *Bubastes* and *Euryspilus*) at the same time as demonstrating significant differences between *Thomassetia* and *Augrabies*-*Jakovleviola*. The sensory formations of the latter bear a certain similarity to *Philanthaxia* and *Aristosoma*. In spite of an obvious resemblance in antennal structures, the problems of status and integration of these taxa remain unsolved until characters other than antennal are thoroughly re-analyzed.

Based on the distinctions in wing venation (the presence of the closed anal cell) *Aristosoma* was contrasted with Thomassetiini (BELLAMY et al. 1987) and placed in Buprestini (BELLAMY 1985), yet its possible affinities to American *Trigonogenium* and *Oaxacanthaxia* BELLAMY, 1991 were maintained (BELLAMY 1991b). Regarding its antennal structure, *Aristosoma* is best matched to *Philanthaxia* and it is quite possible that this genus may produce its own group within Philanthaxiina.

The generic composition of Philanthaxiina remains unclear; as well as *Philanthaxia* it is apparently composed of *Karenaxia* BÍLÝ, 1993 and *Ceylonaxia* BÍLÝ, 1993 (BÍLÝ 1993a,b). However, their antennae were not examined. *Kurosawaia* was separated from *Philanthaxia* (TOYAMA & OHMOMO 1985). BÍLÝ (1993a) indicated its diagnostic characters, such as a simple pronotal punctation, bispinose anal sternite, distinct elytral pubescence, and antennae serrate from the 3rd antennomere. Its antennal organs also differ from those in *Philanthaxia* which, however, does not make impossible their close relationship. Until additional data become available I retain *Kurosawaia* as a separate group of Philanthaxiina.

Tribe Kisanthobiini was established for the monotypic Palaearctic genus *Kisanthobia* by RICHTER (1949) and SCHAEFER (1949) simultaneously but the authorship should be credited to Richter (VOLKOVITSH 1997). HOŁYNSKI (1988, 1993) treated it as a subtribe of Anthaxiini. Indeed, the antennal structure of *Kisanthobia* is very similar to that in *Anthaxia*, differing from previous groups in antennomere shape and position of apical fossae. However, the structure of clypeus and elytral epipleura, as well as some other adult and larval (ALEXEEV & SOLDATOVA 1979, VOLKOVITSH & HAWKESWOOD 1987, 1993, 1994) characters contradicted this placement. It may be supposed that Kisanthobiini are related to Philanthaxiina in spite of some differences in antennal and other structures (for example, well developed prosternal lobe).

The relationships of the monotypic South American tribe Trigonogenini remain obscure. COBOS (1956b) suggested that it may be related to Melanophilini and Kisanthobiini. HOŁYNSKI (1988, 1993) downgraded Trigonogenini to a subtribe of Anthaxiini. Bellamy noted some resemblance of *Trigonogenium* to South African *Aristosoma* of Buprestini (BELLAMY et al. 1987) and also to Mesoamerican *Oaxacanthaxia* (BELLAMY 1991b). The primitive sensory organs as well as other characters prevent the attribution of Trigonogenini to a definite branch of Anthaxioid lineage and the preservation of its tribal rank seems to be a worthwhile decision.

THÉRY (1929) placed *Coomaniella* in Anthaxiini next to *Philanthaxia*. BÍLÝ (1974) elevated it to tribal rank and proposed its affinities with Kisanthobiini and Melanophilini, although he indicated that based on widened antennae and bearing sensory organs from 6th antennomere onwards, eye configuration, body size and coloration sharply dimorphed, and tarsal claws appendiculate, Coomaniellini differs greatly from all the known taxa and demonstrate high specialisation. HOŁYNSKI (1988, 1993) downgraded its rank to subtribe and placed Coomaniellina into Anthaxiini. Sensory organs of *Coomaniella* differ significantly from those in Anthaxiini and Melanophilini, bearing a certain resemblance to *Nascioides*, some species of which also show sharp dimorphism in body shape and coloration (WILLIAMS 1987), and *Kisanthobia*. The closest relationships of Coomaniellini are still an open question.

It seems that Australian taxa usually attributed to Anthaxiini or Melanophilini (BELLAMY 1985) form several small, separated groups like Neocuridina HOŁYNSKI (1988). This assumption is mainly based on the sharp distinctions in larval characters of *Neocuris*, *Anilara*, and *Melobasis* (VOLKOVITSH & HAWKESWOOD 1987, 1993, 1994). Antennal organ states in these taxa produce a morphocline combining the features of both Anthaxiini and Melanophilini that does not help to resolve the problem. So the affinities, position and status of these groups remain uncertain until other characters can be carefully re-analysed.

Curis and *Neocuropsis* (= ?*Curis*) were previously attributed to Stigmoderini (BELLAMY 1985). HOŁYNSKI (1988) established the subtribe Curidina of Anthaxiini and then placed in it Neotropical *Cylindrophora* and *Anthaxioides* COBOS, 1976, unknown to me (HOŁYNSKI 1989). In doing so, he divided *Cylindrophora*, retaining there only a species part, whereas another species group was transferred to subgenus *Curis* (*Ctenoderus* GERMAIN, 1856) and a third one to newly erected subgenus *Anthaxia* (*Bilyaxia* HOŁYNSKI, 1989) of Anthaxiina. However, the sensory organs of *Curis* and *Neocuropsis* are much closer to *Neobubastes* and *Eububastes* than to *Cylindrophora* or *Anthaxia*, making their affinities to Anthaxiini rather problematic.

The apical organs of *Cylindrophora* (including *Ctenoderus* and *Bilyaxia*) are represented with closed or partly open cavities, the latter being found in *C.* (*Ctenoderus*) *maulica* and *C.* (s.str.) *verecunda*. Such an antennal structure does not support integration of *Cylindrophora* with *Curis* in Curidina (HOŁYNSKI 1989). On the other hand, the similarity in wing venation and male genitalia (COBOS 1956a) confirms the relationships of *Cylindrophora* s.str. and *Bilyaxia* to Anthaxiina. The head structure of *Ctenoderus* does not exclude affinities to Australian Stigmoderini, *Curis*, and *Neocuris* and supports the possible polyphyly of *Cylindrophora* as HOŁYNSKI (1989) has suggested. It is also quite possible that *Cylindrophora*, being a monophyletic taxon, forms a separate South American group of Anthaxiini together with *Tetragonoschema* and *Agrilaxia* KERREMANS, 1903.

Nearly all the Old World and Nearctic Anthaxiini studied demonstrate a great similarity in antennal (except *Brachanthaxia*) and larval (BÍLÝ 1975, SOLDATOVA 1970, 1973, VOLKOVITSH & HAWKESWOOD 1987, 1993, 1994, VOLKOVITSH & BÍLÝ 1997) characters. Their complete identity in *Chalcogenia* and *Anthaxia* supported by other characters allowed the transfer of *Chalcogenia* from Melanophilini to Anthaxiini (VOLKOVITSH & BÍLÝ 1997) in spite of distinctions in pronotal sculpture, regarded as a key character to distinguish these tribes. As noted above, the characteristic pathway of apical cavity formation in African *Brachanthaxia*, differing from that in Australian and Neotropical taxa, rather reflects the independent evolution of Old World and Nearctic Anthaxiini, so these can be separated as subtribe Anthaxiina in contrast to the mainly Neotropical *Cylindrophora* group.

The American tribe Xenorhipini was established by COBOS (1986a). HOŁYNSKI (1988) being apparently unaware of Cobos' publication, repeatedly erected Xenorhipina as subtribe of Anthaxiini. The most characteristic trait of this group is a sharp dimorphism: the males have strongly modified pectinate antennae whereas the females preserve typical buprestoid organs differing from those in Anthaxiini. Moreover, there are marked distinctions in antennal structure between *Xenorhipis* and *Hesperorhipis* on the one hand, and *Trichinorhipis* on the other. The larvae of *Xenorhipis* and *Hesperorhipis* (that of *Trichinorhipis* is unknown) share with those of Anthaxiini the similar pronotal grooves, glabrous prothoracic plates, and the position of antennae, but they differ in other important characters such as the absence of double lateral ambulatory pads on metathorax, the hypertrophy of the 1st and nearly complete reduction of the 2nd antennal segment, and the presence of microspinulated areas on the mouthparts (VOLKOVITSH, unpublished data). Thus both antennal and larval characters support the elevation of Xenorhipini as a separate tribe of Anthaxioid lineage. The distinctions in antennal structures between *Xenorhipis*-*Hesperorhipis* and *Trichinorhipis* also suggest the separation of them into two generic groups or subtribes, although their relationships and status invite further investigation.

The assumption about the close affinities and possible common ancestry of Melanophilini and Chrysobothrini proposed by HOŁYNSKI (1993) is supported by great similarity in antennal and larval characters.

Antennal and larval characters of *Chalcogenia* and Australian genera indicated that these must be removed from Melanophilini in spite of similar pronotal sculpture. On the other hand, Nearctic *Juniperella* which is usually placed in Buprestini (BELLAMY 1985), on the basis of antennal structure and some other characters including larval ones (VOLKOVITSH, unpublished data) should be transferred to Melanophilini. The characteristic apical (open cavities) and lateral (extensive indifferentiated fields) sensory organs suggest that *Juniperella* may be treated as a separate and very primitive group of Melanophilini.

The position of Australian *Merimna* remains a mystery. Some authors have attributed it to Melanophilini (KERREMANS 1903, COBOS 1986a,b), others to Actenodini (BELLAMY 1985, 1986, HOŁYNSKI 1993). In regard to its antennal structure *Merimna* is much closer to Melanophilini, more especially to *Melanophila* than to Actenodini. Yet, based on the similarity in sensory organs, its possible relation to Australian Anthaxioid taxa (e.g. *Melobasis*) must not be ruled out. I presume that *Merimna* should be elevated to its own subtribe or even tribe whose affinities can be properly evaluated only by way of re-examining other characters.

COBOS (1986a,b) placed *Cromophila* in Melanophilini although he noted some differences in antennal structures. Its sensory organs (round apical fossae instead of cavities, the presence of lateral depressions) differ greatly from those in Melanophilini, demonstrating a certain similarity to Actenodini. It is not unlikely that such a state is an initial one for both Actenodini and Chrysobothrini, and that *Cromophila* should be placed in a separate subtribe of Actenodini or even in its own tribe of Chrysobothrinioid branch.

As well as in its peculiar tarsomere structure, Actenodini also differs from Chrysobothrini in having apical organs that are always open. On the other hand, the antennal structures of Actenodini show a significant variability from states similar to

Cromophila to ones resembling Chrysobothrini, even within the same genus. Sharp distinctions in the antennal structures of *Actenodes* prove its possible polyphyly, e.g. the sensory organs of *A. costipennis* LAP. & GORY and *A. chalibaeitarsis* CHEVR. are much closer to those in *Belionota* than to other *Actenodes* species. Antennal organs within a further group (*A. bellula* Mannh., *A. auronotata* LAP. & GORY, *A. calcarata* CHEVR.) resemble those in Agaeocerina. The third group (*A. goryi* Mannh., *A. nobilis* L., *A. adonis* LAP. & GORY.) has much in common with Chrysobothrini owing to its geniculate antennae.

Chrysobothrini possess the most specialised antennae among Buprestidae the structure of which, in contrast to Actenodini, is quite uniform, being characterised by geniculate shape and apical cavities completely closed in the first place. There is no question that this structure originated from the states inherent in some *Actenodes* groups.

Incertae sedis

Fahraeusia, originally established as a distinct genus, was subsequently downgraded to a subgenus of *Oedisterna* (Psilopterini) (see BELLAMY 1985). Based on antennal structure and wing venation TOYAMA (1987) restored its generic level and transferred *Fahraeusia* to Buprestini close to South American taxa, meaning apparently those that I placed in Hypoprasini (Chalcophorinae). In fact, the sensory organs of *Fahraeusia* being of buprestoid type have nothing in common either with those in Psilopterini, or Hypoprasini, most resembling African *Thomassetia*. However, it is quite possible that the sensory organs of *Fahraeusia* actually resulted from modification as takes place in *Saundersina*, *Dicerca*, and some others where antennal structure differs greatly from that of closely related taxa. The similarity in locality of shortened U1 sensillae on the top of the apical antennomere provides reason enough to suggest that *Fahraeusia* may belong to *Chalcoplia* group of Chalcophorinae and should be placed there next to *Embrikillium*.

Galbelloid complex (Galbellinae)

Antennal structures of *Galbella* are in agreement with primitive states found in some representatives of Polycestoid and Agriloid complexes but its larval characters (the structure of antennae, mouthparts and spiracles) are of Buprestoid type, which never occurs in Agriloid taxa (VOLKOVITSH, unpublished data). This counteridicates the placement of *Galbella* in Trachyini (HOŁYNSKI 1993). The larvae of *Galbella* demonstrate a number of autapomorhies and, additionally, they are xylophages in contrast to the mining larvae of Trachyini. In my opinion *Galbella* occupy an isolated position and until new data is available, Galbellinae should hold the rank of subfamily as suggested by REITTER (1911) and COBOS (1986a).

Summary

Based on peculiarities of antennal structure genesis, the main phylogenetic stocks, lineages, branches and groups in Buprestidae are recognised as follows: Schizopoid complex (Schizopodinae): Schizopoid lineage (Schizopodini), Dystaxioid lineage (Dystaxiini THÉRY, 1929); Julodoid complex (Julodinae), no subordinate

categories established; Polycestoid complex (Polycestinae): Acmaeoderioid lineage (Mastogeniini Le Conte & Horn, 1883, Acmaeoderini Kerremans, 1893, Ptosimini Kerremans, 1902, Paratrachyini Cobos, 1980), Polyctesioid lineage (Thrincopygini Le Conte, 1861, *Chrysophana* Le Conte, 1860 group, Polyctesini Cobos, 1955), Polycestioid lineage (Bulina Bellamy, 1995, Polycestini Lacordaire, 1857, Tyndarini Cobos, 1955), Xyroscelini Cobos, 1955, Prospherini Cobos, 1980, and Astraeusini Cobos, 1980 as separate groups; Chalcophoroid complex (Chalcophorinae): Chalcophorioid lineage (Vadonaxiini Descarpentries, 1969, Paratassini Bílý & Volkovitsh, 1996, *Nanularia* Casey, 1909 generic group, *Chalcoplia* Saunders, 1871 generic group, Chalcophorini including Chrysochroina Laporte, 1835, Evidini Toyama, 1987, Hypoprasini Hołynski, 1993, **stat.nov.**, Poecilonotini Alexeev & Bebka, 1970), Psilopterioid lineage (Sphenopterini Lacordaire, 1857, Psilopterini Lacordaire, 1857 including Hippomelanina Hołynski, 1993, Pseudoperotina Toyama, 1987, and Haplotrinchina Hołynski, 1993); Buprestoid complex (Buprestinae): Mendizabalini Cobos, 1968, *Philandia* Kerremans, 1907 generic group, Maoraxiini Hołynski, 1984 as isolated groups, Buprestioid lineage (Julodimorphini Kerremans, 1902; Buprestinioid branch: Nascionina Hołynski, 1988, *Pygicera* Kerremans, 1903 generic group, Epistomentini Levey, 1978, *Neobuprestis* Kerremans, 1903 generic group, Buprestini including Trachykelina Hołynski, 1988, Lamprocheilina Hołynski, 1993 and Agaeocerina Nelson, 1981, Exagistini Toyama, 1987 and Phrixini Cobos, 1975; Stigmoderinioid branch: *Buprestina* Obenberger, 1923 generic group, Bubastini Obenberger, 1920, *Microcastalia* Heller, 1891 generic group, Stigmoderini Lacordaire, 1857; *Aglaostola* branch: *Aglaostola* Saunders, 1871, *Hilarotes* Saunders, 1871, *Spectralia* Casey, 1909 generic groups; *Pterobothris* Fairmaire & Germain, 1858 branch: Pterobothrini **trib.nov.**), Anthaxioid lineage (Thomassetiinioid branch: *Neobubastes* Blackburn, 1892 generic group, Thomassetiini Bellamy, d'Hotman & Holm, 1987, *Aristosoma* Saunders, 1871 generic group, Philanthaxiina Hołynski, 1988 including *Kurosawaia* Toyama & Ohmomo generic group, 1985, Kisanthobiini Richter, 1949; Anthaxiinioid branch: Trigonogenini Cobos, 1956, Coomaniellini Bílý, 1974, Curidina Hołynski, 1988, Neocuridina Hołynski, 1988, *Torresita* Gemminger & Harold, 1869, *Theryaxia* Carter, 1928, *Anilara* Saunders, 1871, *Melobasis* Laporte & Gory, 1837 generic groups, Anthaxiini Laporte & Gory, 1837, Xenorhipini Cobos, 1986), Chrysobothrioid lineage (Melanophilinioid branch: Melanophilini Bedel, 1921, *Merimna* Saunders, 1871 generic group; Chrysobothrinioid branch: *Cromophila* Cobos, 1986 generic group, Actenodini Kerremans, 1893, Chrysobothrini Laporte & Gory, 1837); Galbelloid complex (Galbellinae), no subordinate categories recognised. These groups do not fully correspond to the currently accepted formal taxa. Based on the antennal structures combined with other adult and larval characters the following taxonomical changes are suggested. Polycestinae, Chalcophorinae, and Galbellinae are reconstituted as distinct subfamilies. Within Polycestinae: Mastogeniini is restored as a distinct tribe instead of a subtribe of Thrincopygini; Odetteina **subtr.nov.** is established for *Odettea* Baudon 1966 and transferred from Polycestina to Acmaeoderini; Ptosimini Kerremans 1902 is regarded to be monotypic; *Acmaeoderoides* Van Dyke 1942 which is removed from Ptosimina, and Nothomorphina Cobos, 1955 comprising only *Nothomorpha* Saunders, 1871 and *Nothomorphoides* Holm, 1986 are transferred from Thrincopygini to Acmaeoderini and regarded as either distinct subtribes or two independent generic groups of Acmaeoderina; *Bulina* is transferred from Tyndarini next to Polycestini as a separate generic group; Tyndarini is treated either as a distinct tribe comprising subtribes Tyndarina, Tylauchenina Cobos, 1973, Acherusina Cobos, 1955, and Pseudacherusina Cobos, 1980, or as a subtribe of Polycestini with the above-mentioned taxa downgraded to generic group level; Thrincopygini is regarded as a monotypic tribe; *Chrysophana* is transferred from Bubastini back to Polycestinae but its previous placement in Polyctesini is not supported and it is separated within its own group; the tribal status and separate positions of Xyroscelini, Prospherini and Astraeusini are restored. Within Chalcophorinae: *Nanularia* and *Ampheremus* Fall, 1917 which are removed from Hippomelanina, and *Chalcoplia*, *Descarpentriesiola* Cobos, 1978, and *Embrikillium* Obenberger, 1936 are placed in their own groups; Chrysochroina is considered to be a subtribe of Chalcophorini, *Rhabdolona* Obenberger, 1924 is transferred to Chalcophorini from Buprestini, and several generic groups are separated; Hypoprasini Hołynski **stat.nov**. upgraded to tribal rank and Euchromatina Hołynski, 1993, Hypoprasina Hołynski, 1993, Pristipterina Hołynski, 1993, Euplectaleciina Hołynski, 1993, and Cinyrina Cobos, 1979, **stat.nov.** transfered to this tribe from Buprestini, *Cordillerita* Obenberger, 1928 is transferred to Hypoprasina from Dicercini Kerremans, 1893; *Poecilonota* Eschscholtz, 1829 and *Palmar* Schaefer, 1949 (as *Lampra*) are placed in Poecilonotini instead of their previous position in Dicercini or Psilopterina, *Nesotrinchus* Obenberger, 1924 is transferred to the former tribe from Haplotrinchina as a separate group; Sphenopterini is transferred from Anthaxiini to Chalcophorinae next to Psilopterini and its tribal rank is restored; Psilopterini is resurrected as a distinct tribe of Chalcophorinae, Hippomelanina, Haplotrinchina, and Pseudoperotina are transferred to this tribe from Buprestini as well, *Chalcopoecila* SAUNDERS 1871 is transferred from Pseudoperotina to Psilopterina, and some generic groups within the latter are suggested. Within Buprestinae: Mendizabalini is believed to be a polyphyletic taxon and *Philandia* is placed in its own group; Maoraxiini are reconstituted as a distinct tribe instead of a subtribe of Anthaxiini; Nascionina is removed from Anthaxiini and regarded as a probable polyphyletic group; *Pygicera* and *Xenocyria* OBENBERGER, 1947 are restored as distinct genera instead of subgenera of *Epistomentis* SOLIER, 1833 and *Cyria* SOLIER, 1833 respectively, *Pygicera* is separated into its own group; *Neobuprestis* which is restored from subgenus of *Microcastalia*, and *Balthasarella* OBENBERGER, 1958 are placed in a separate group; Trachykelina and Agaeocerina are transferred from Anthaxiini and Thrincopygini respectively to Buprestini; the tribal level of Exagistini and Phrixini is restored, the latter is regarded to made up of *Paraphrixia* SAUNDERS 1871 and *Phrixia* DEYROLLE, 1864 exclusively; *Buprestina* and *Microcastalia* are separated into their own groups behind Bubastini and Stigmoderini, Bubastini is regarded as comprising *Euryspilus* LACORDAIRE, 1857 and *Bubastes* LAPORTE & GORY, 1836; *Castiarina* GORY & LAPORTE, 1838 (*Hypostigmodera* BLACKBURN, 1892) is suggested as a distinct genus; an affinity of *Aglaostola*, *Hilarotes*, *Spectralia*, and *Ditriaena* WATERHOUSE, 1911 is assumed; it is suggested that *Pterobothris* be placed in its own tribe, *Pterobothrini,* **trib.nov.**; *Neobubastes* and *Eububastes* OBENBERGER, 1928 are removed from Bubastini and placed in their own group; an affinity of *Aristosoma* to Philanthaxiina instead of its present placement in Buprestini is proposed; *Juniperella* KNULL 1947 is transferred from Buprestini to Melanophilini.

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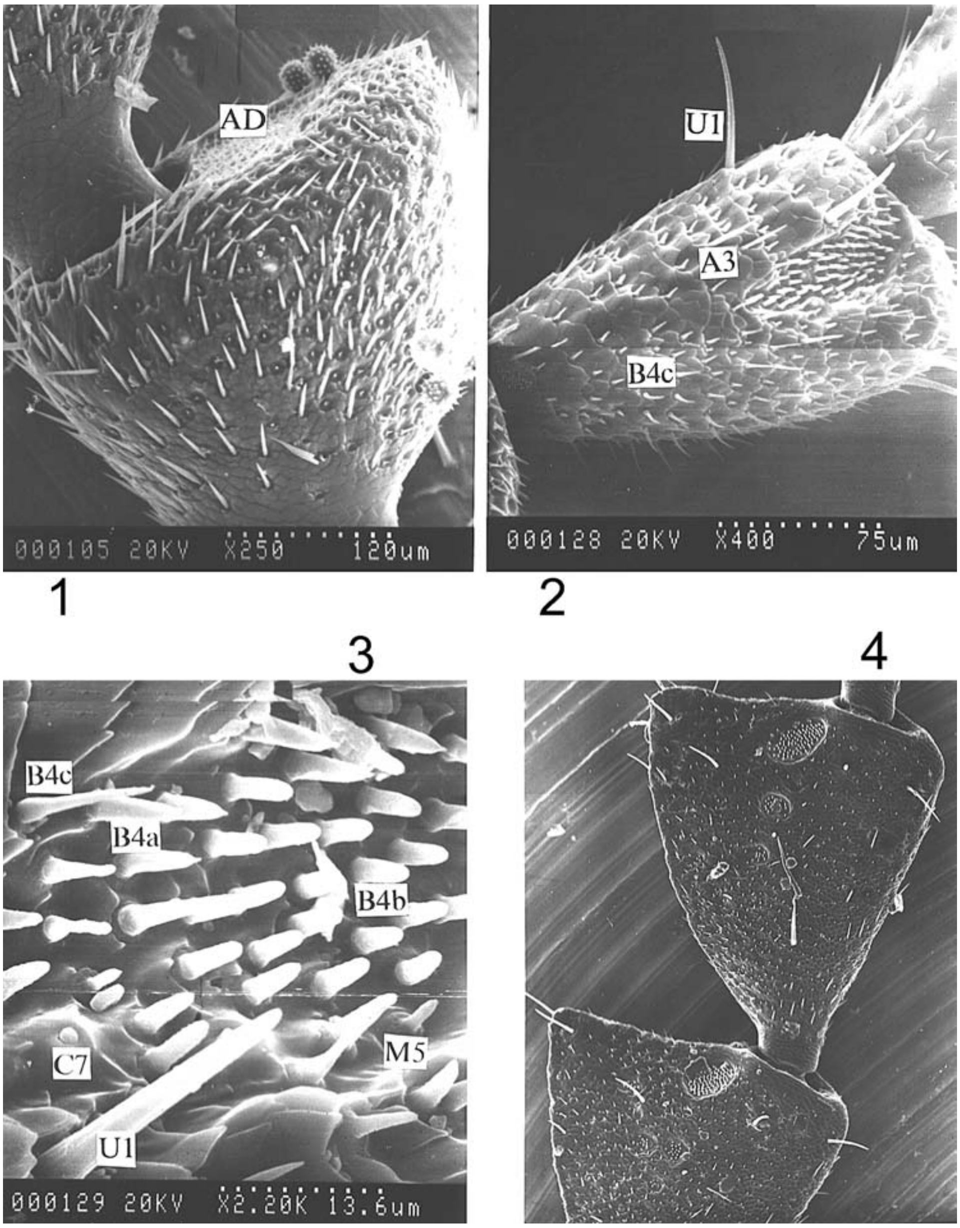
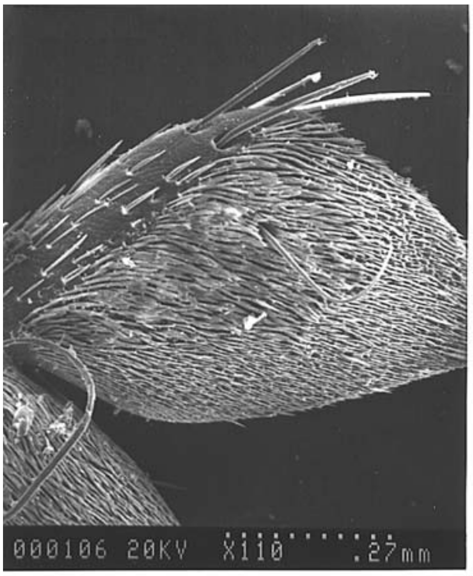


Figs 1–4: *Schizopus*, *Glyptoscelimorpha*, *Dystaxia*. – 1. *Schizopus laetus*: male, 7th, internal, showing apical depression (AD) and external B4c and A3 sensillae, ×250; 2, 3. *Glyptoscelimorpha viridis*: male; 2: 8th, internal, showing subapical depression comprising internal sensillae and external uniporous (U1), basiconic (B4c), and aporous (A3) sensillae, ×400; 3: 5th, internal, showing lower part of subapical depression comprising internal basiconic (B4a, B4b), multiporous (M5) and campaniform (C7) sensillae, and ajacent external uniporous (U1) and basiconic (B4c) sensillae, ×2200; 4. *Dystaxia elegans*: 8–9th, internal, showing subapical and lateral pits and fossae, ×100.



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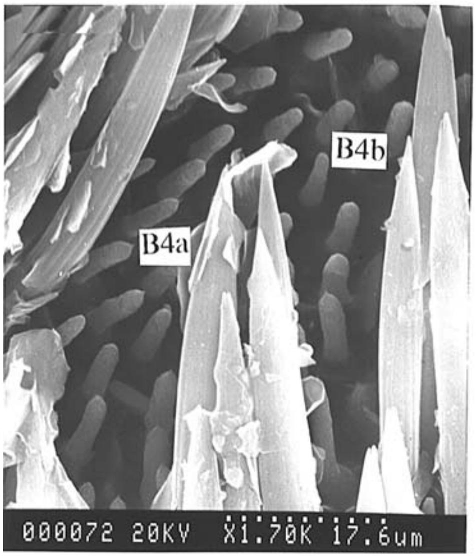

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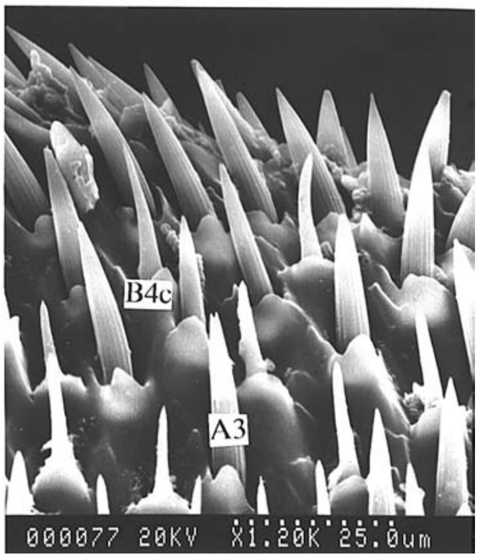
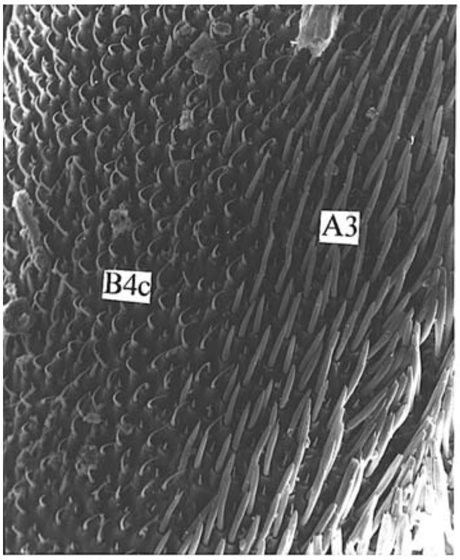


Figs 5–8: *Julodis*, *Aaata*. – 5. *Julodis variolaris*: female, 11th, internal, ×110; 6–8. *Aaata finchi*, male, 11th, internal; 6: side near an outer margin, showing sensory zones (SZ) and sensillar pits (SP), ×2200; 7: sensory zone comprising internal basiconic B4a and B4b sensillae, ×1700; 8: sensory field at anterior margin comprising shortened modified aporous (A3) and basiconic (B4c) sensillae, ×1200.



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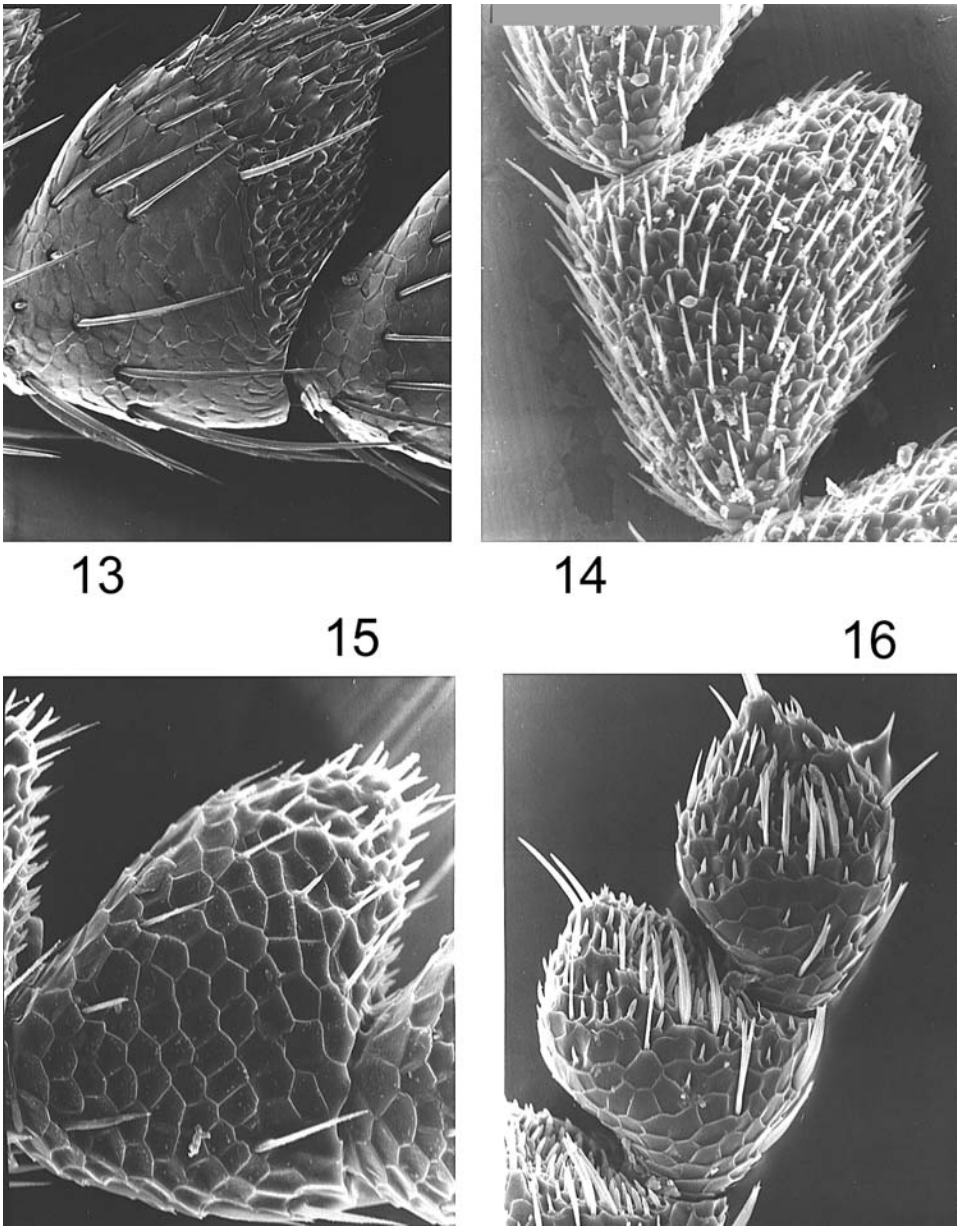
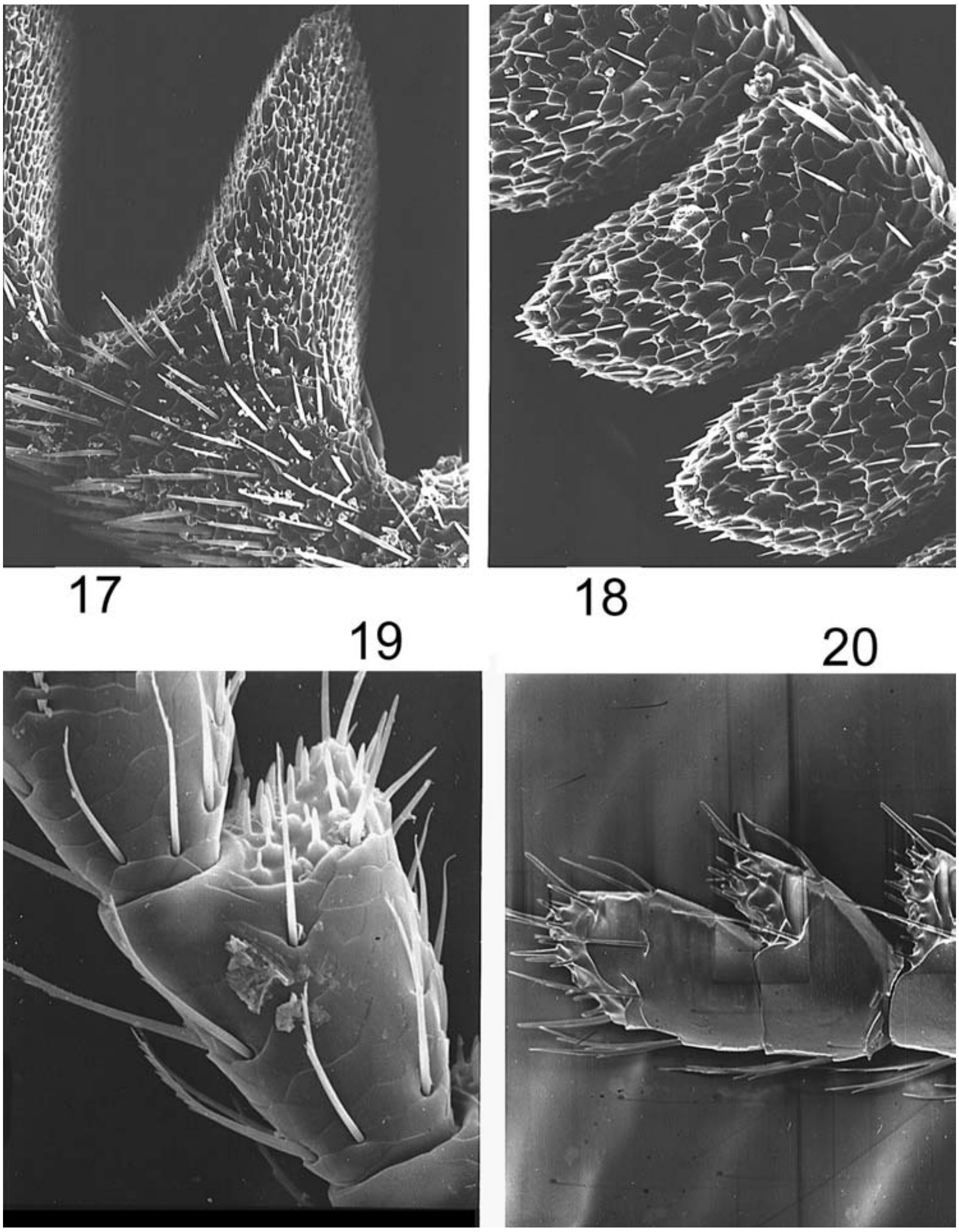
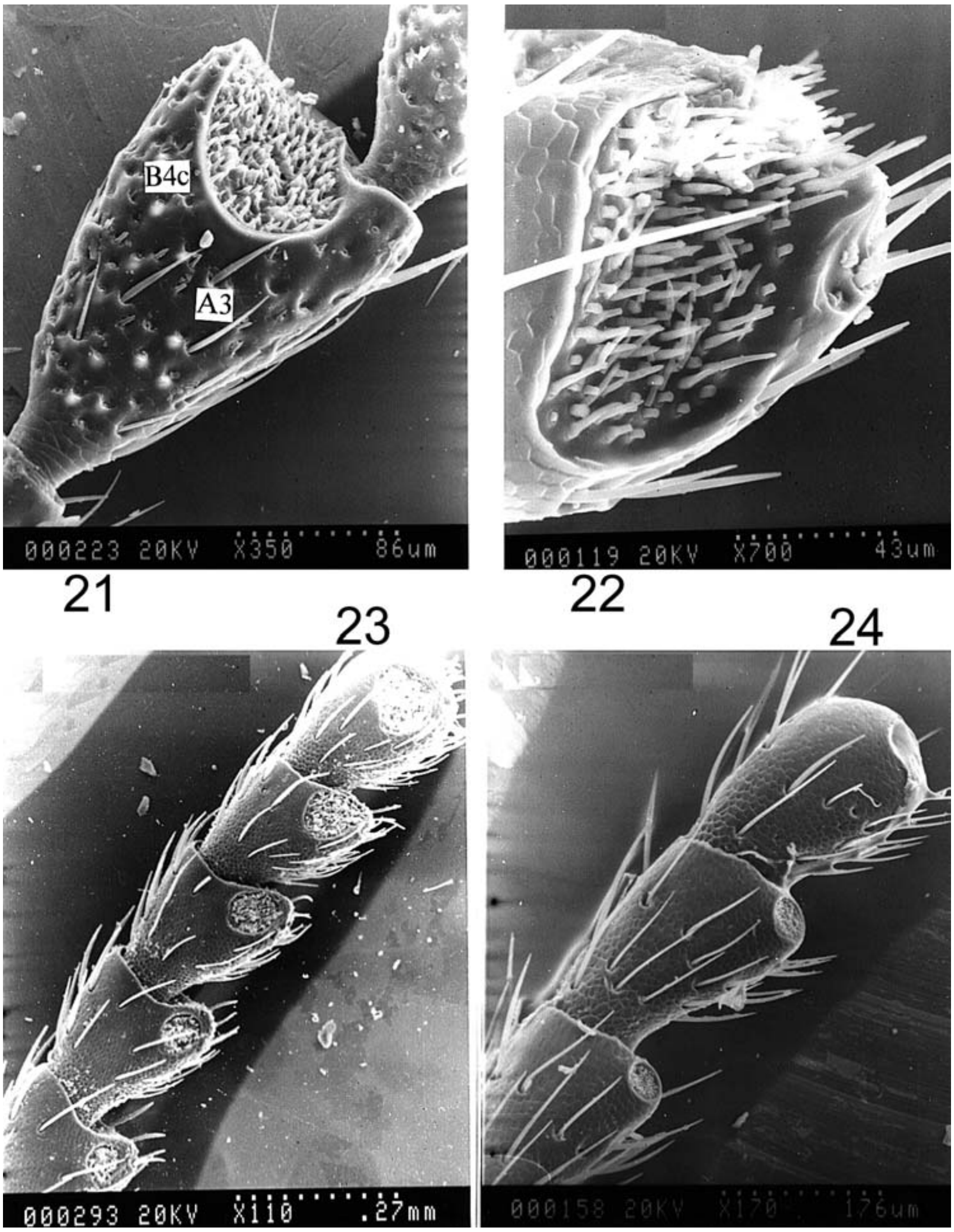
Figs 9–12: *Sternocera*, *Mastogenius*, *Odettea*. – 9. *Sternocera orissa luctifera*: 8th, internal, showing fields of modified aporous (A3) and unguiculate basiconic (B4c) sensillae, ×300; 10. *Mastogenius cyaneus*: female, 10–11th, internal, showing sensory zones at apical corners and dispersed uniporous (U1) and aporous (A3) sensillae, ×700; 11. *M. robustus*: male, 9–11th, internal, ×250; 12. *Odettea laosensis*: female, 8–11th, internal, ×300.



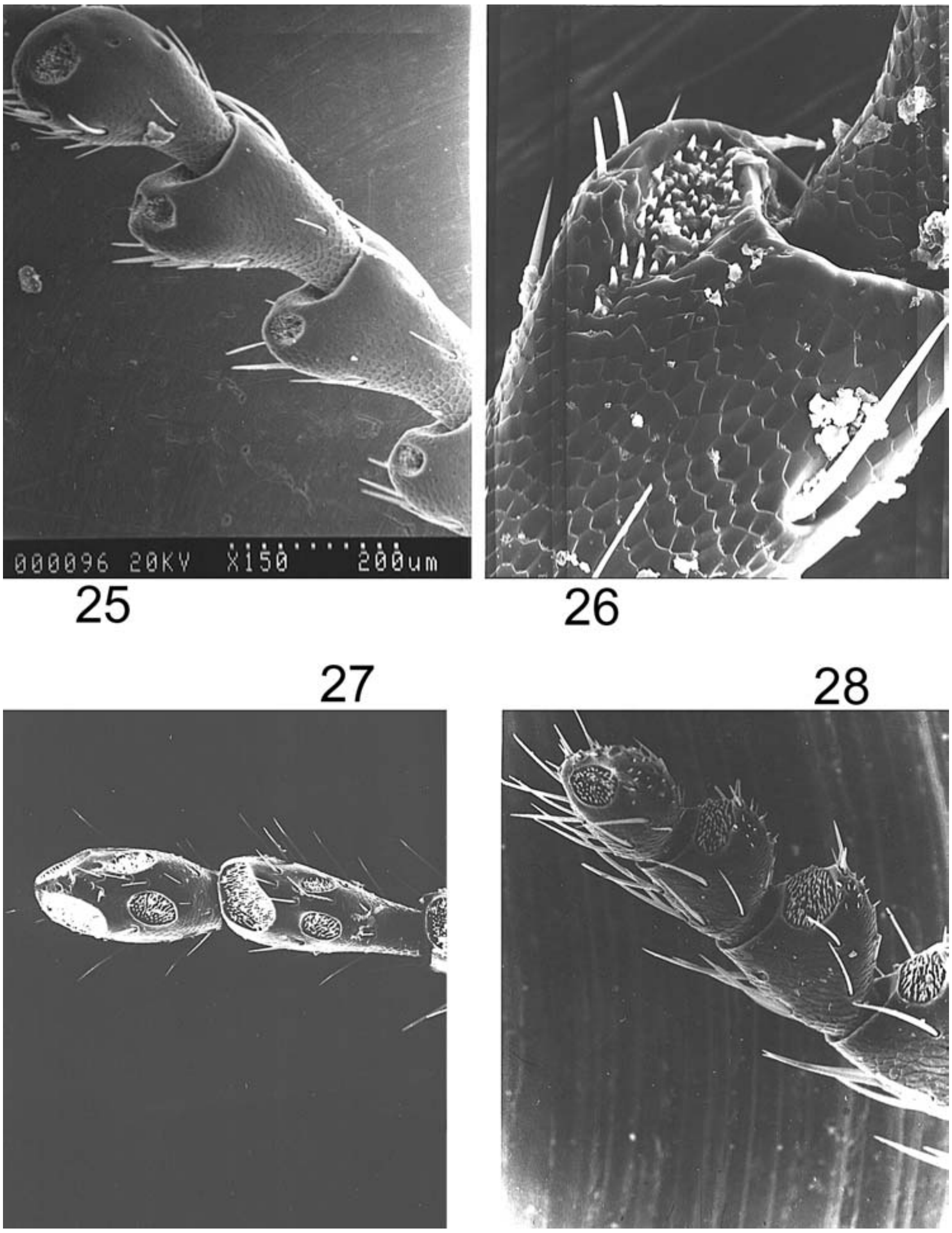
Figs 13–16: *Ptosima*, *Nothomorphoides*, *Nothomorpha*, *Acmaeoderopsis*. – 13. *Ptosima undecimmaculata*: 10th, internal, ×450; 14. *Nothomorphoides irishi*: male, 10th, internal, ×400; 15. *Nothomorpha major*: 10th, external, ×700; 16. *Acmaeoderopsis jaguarina*: 10–11th, internal, ×450.



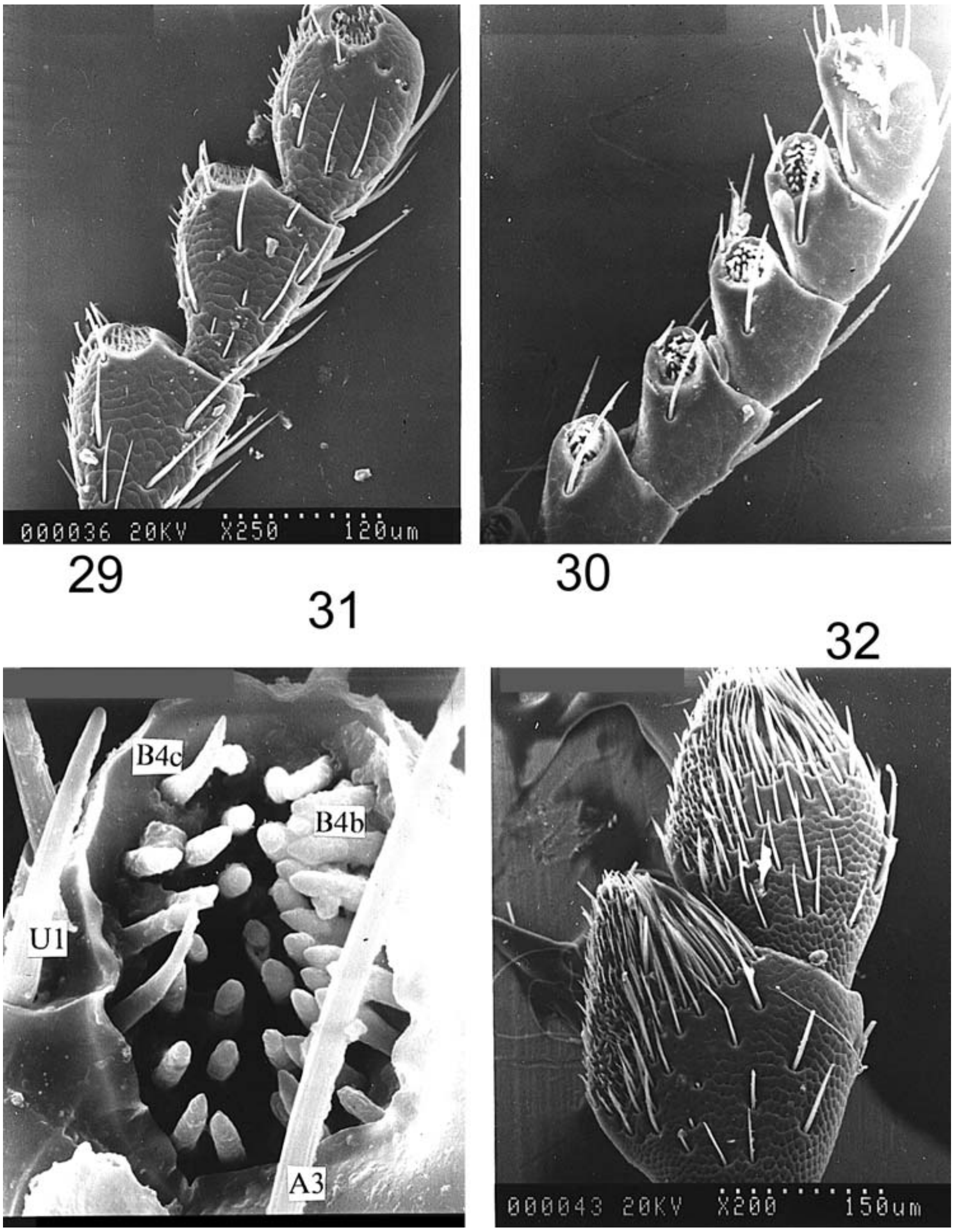
Figs 17–20: *Acmaeoderella*, *Sponsor*, *Paratrachys*. – 17, 18. *Acmaeoderella* (*Euacmaeoderella*) *subcyanea*, 17: male, 9th, internal, ×300; 18: female, 9th, internal, ×450; 19. *Sponsor* (s.str.) *emmae*: 8th, internal, ×800; 20. *Paratrachys hederae*: 9–11th, internal, ×700.



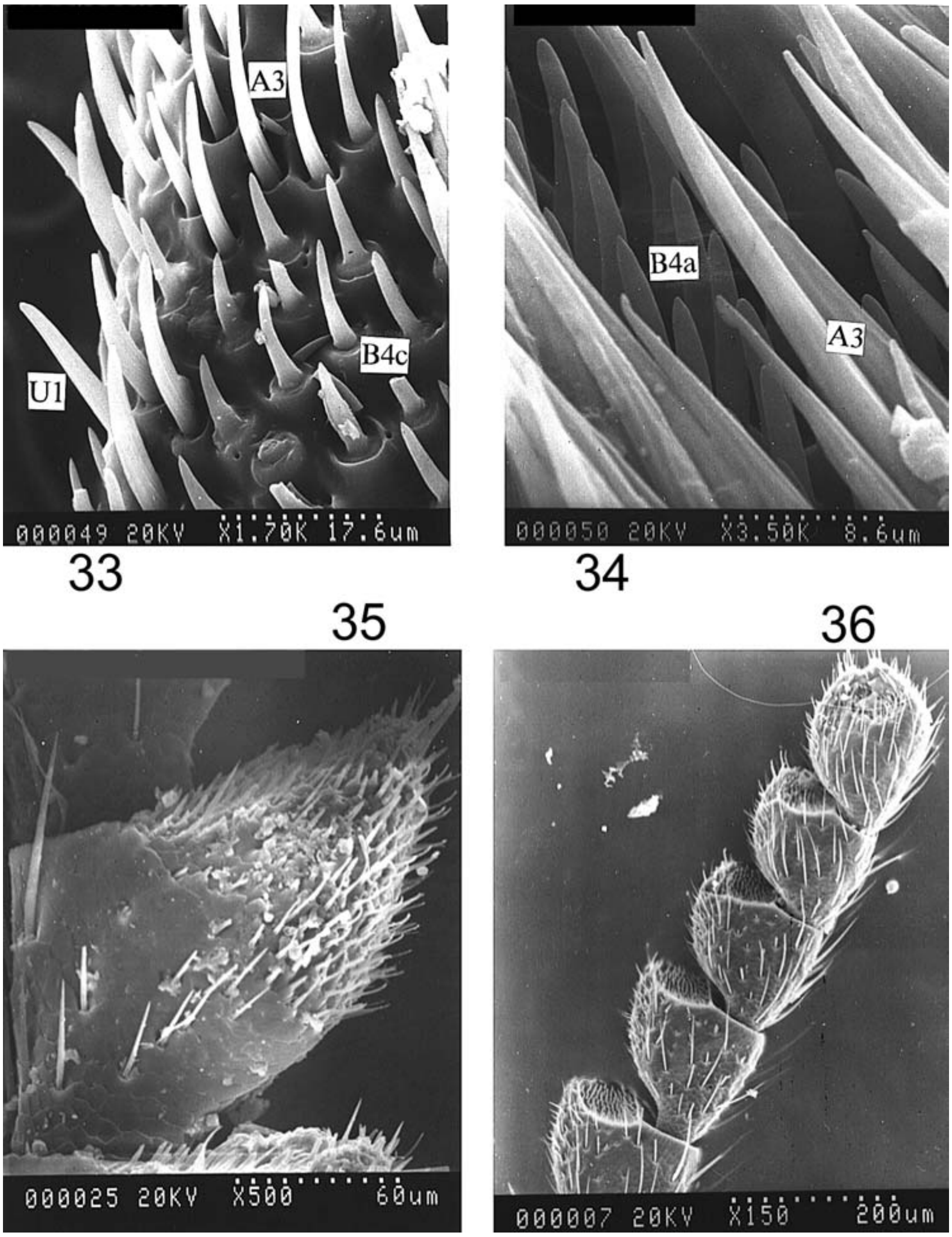
Figs 21–24: *Bulis*, *Jelinekia*, *Paracastalia*, *Thurntaxisia*. – 21. *Bulis bivittata*: 8th, internal, showing subapical fossae and scattered basiconic (B4c) and aporous (A3) sensillae, ×350; 22. *Jelinekia barri*: male, 10th, internal, ×700; 23. *Paracastalia ornatipennis*: 7–11th, internal, ×110; 24. *Thurntaxisia alexandri*: 9–11th, internal, ×170.



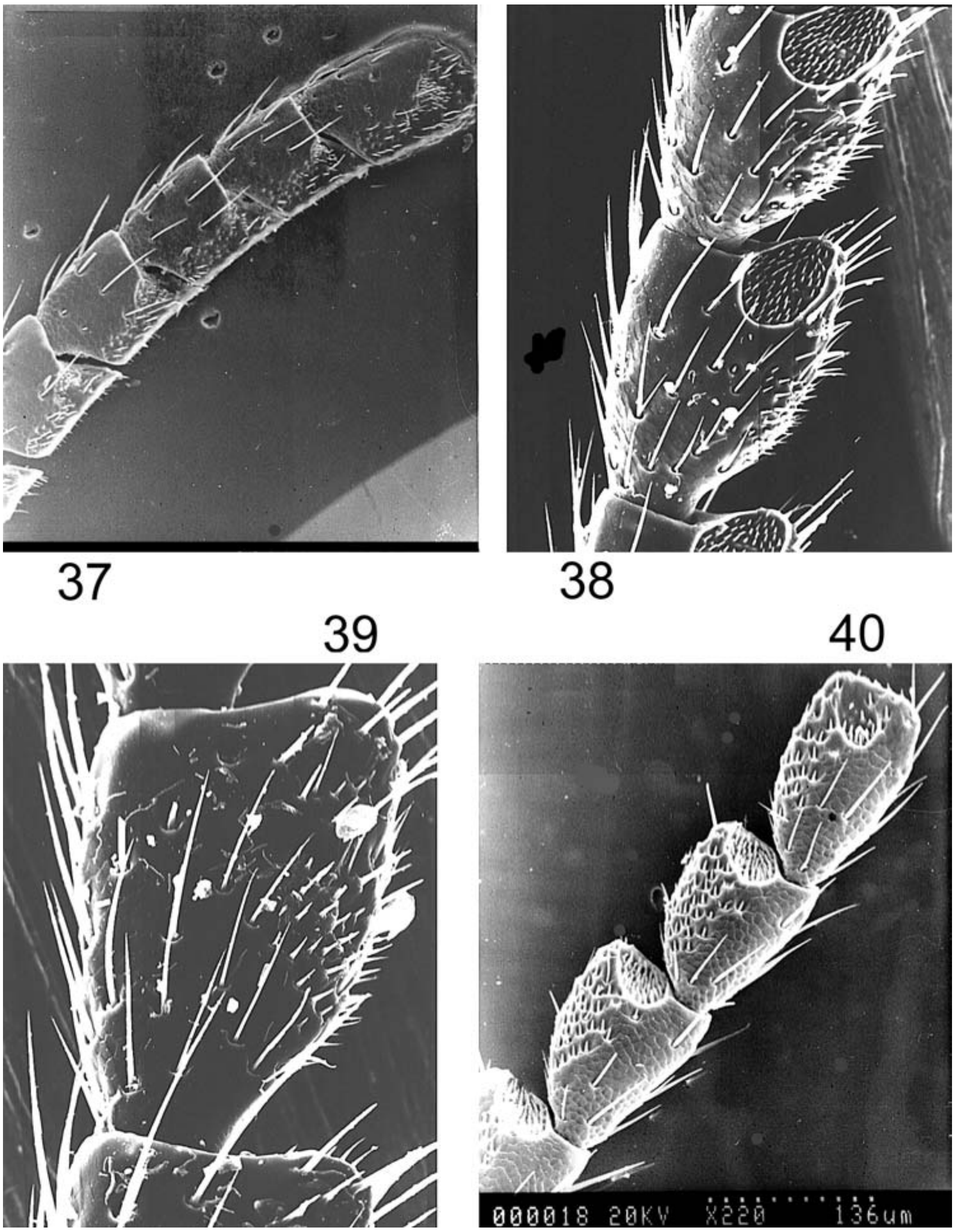
Figs 25–28: *Pseudocastalia*, *Strigopteroides*, *Polycesta*, *Tylauchenia.* – 25. *Pseudocastalia arabica*: 8–11th, internal, showing confluenting internal and external fossae on 10th, ×150; 26. *Strigopteroides aegyptiacus*: 6th, internal, ×450; 27. *Polycesta porcata*: 10–11th, inner margin, ×100; 28. *Tylauchenia crassicollis*: 8–11th, internal, ×150.



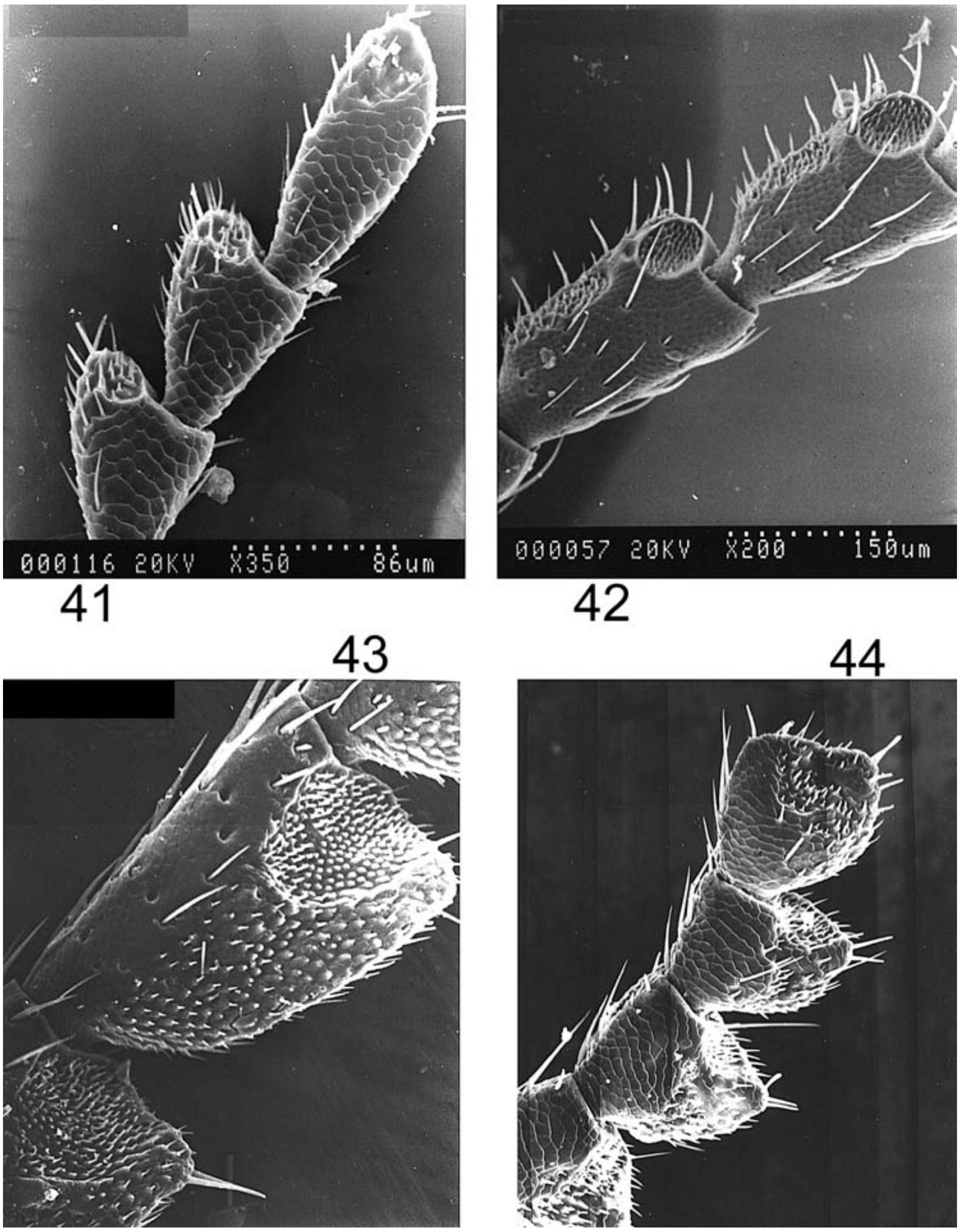
Figs 29–32: *Tyndaris*, *Acherusia*, *Pseudacherusia, Thrincopyge*. – 29. *Tyndaris* (s.str.) *planata*: 9–11th, internal, ×250; 30, 31. *Pseudacherusia bartoni*: internal; 30: 7–11th, ×300; 31: 10th, showing apical fossa comprising internal basiconic (B4b and B4c) and external uniporous (U1) and aporous (A3) sensillae, ×2500; 32. *Thrincopyge ambiens*: male, 10–11th, internal, ×200.



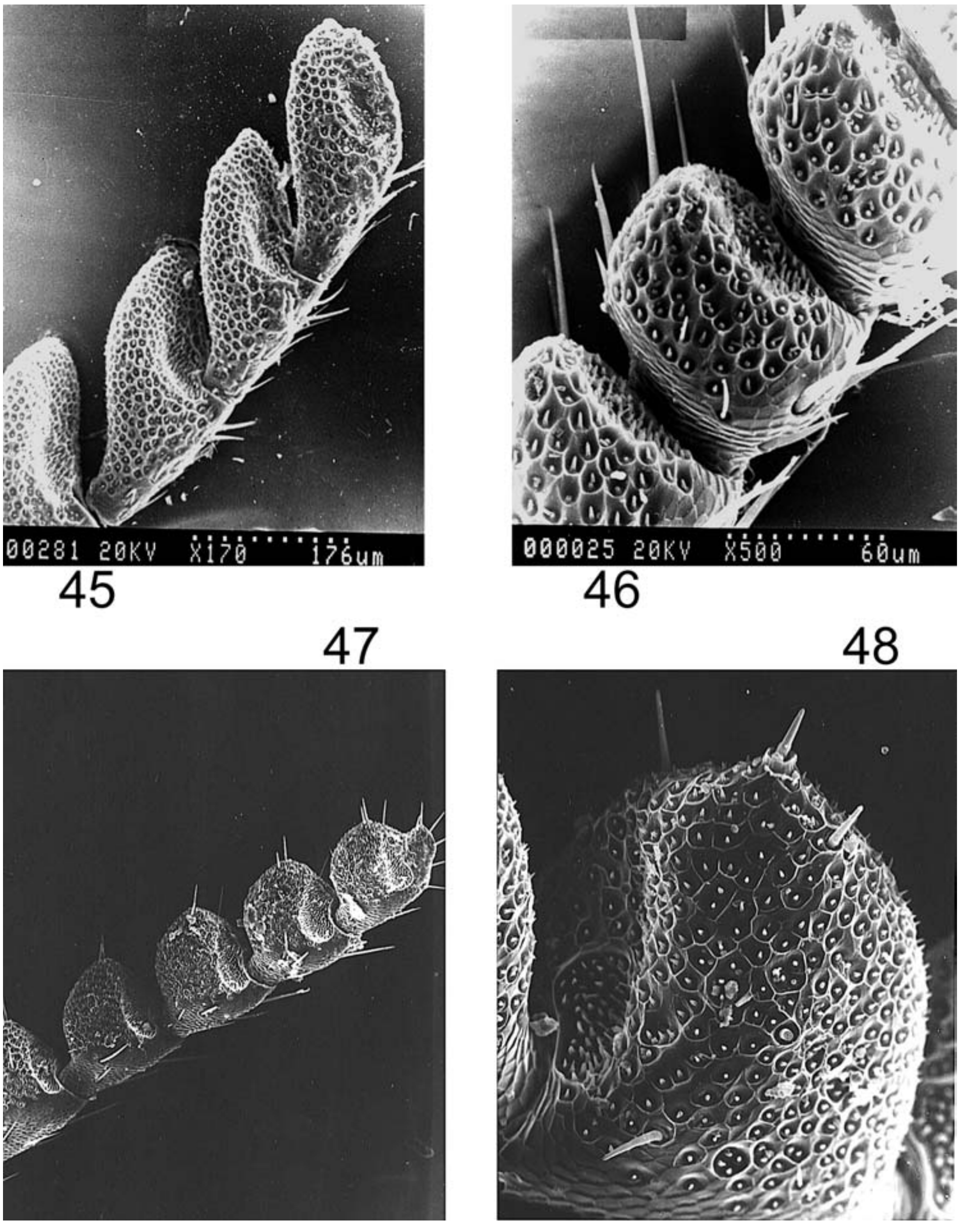
Figs 33–36: *Thrincopyge*, *Schoutedeniastes*, *Bellamyina*. – 33, 34. *Thrincopyge ambiens*: male, internal; 33: 9th, showing field of external uniporous (U1), aporous (A3), and basiconic (B4c) sensillae, ×1700; 34: 9th, showing subapical sensory zone of basiconic (B4a) sensillae concealed by external aporous (A3) sensillae, ×3500; 35. *Schoutedeniastes amabilis*: 8th, internal, ×500; 36. *Bellamyina hunanensis*: 7– 11th, internal, ×150.



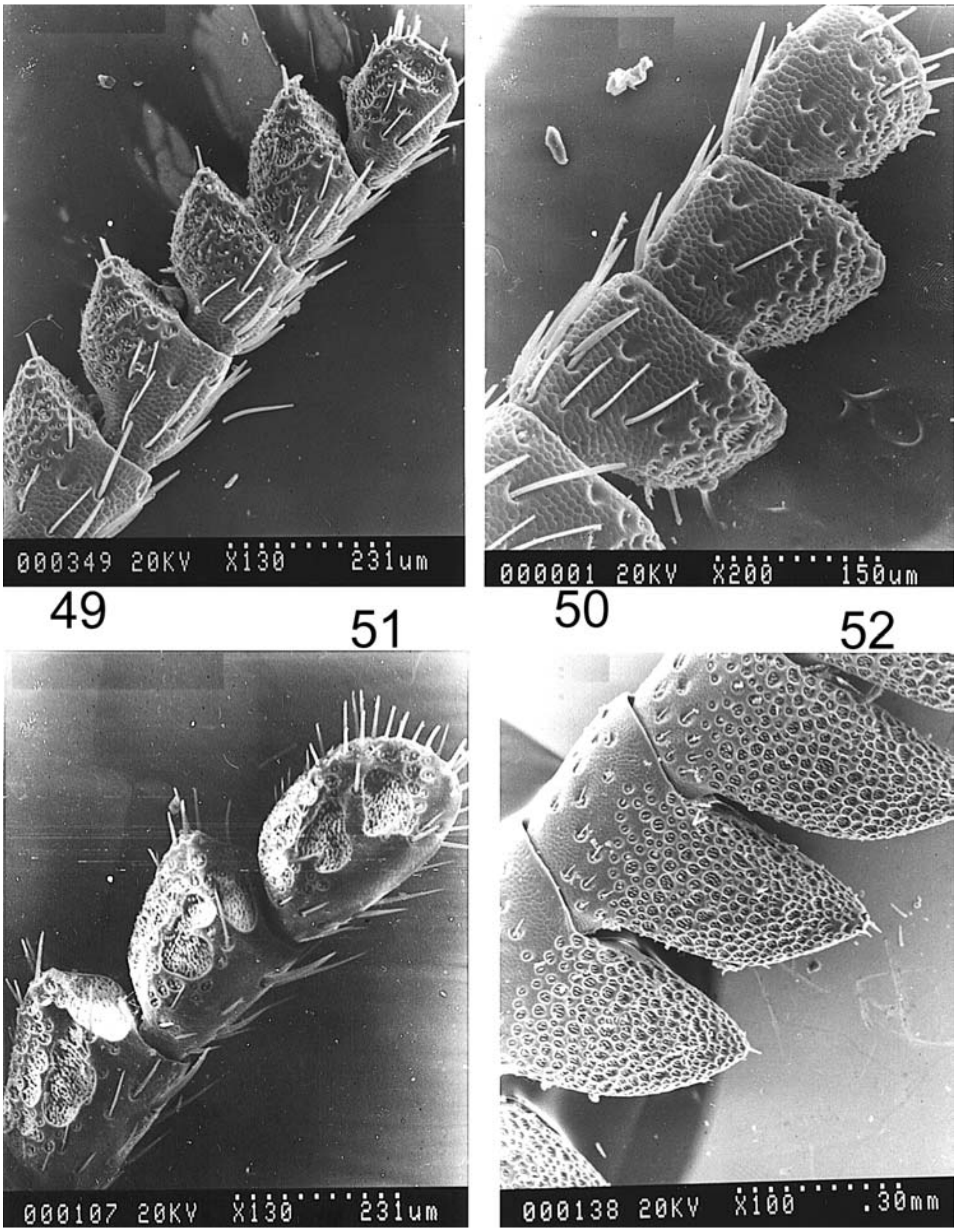
Figs 37–40: *Paraxenopsis*, *Polyctesis*, *Chrysophana*. – 37. *Paraxenopsis* sp.: female, 7– 11th, internal, ×120; 38, 39. *Polyctesis rhois,* 38: 6–7th, internal, ×200; 39: 6th, external, ×300; 40. *Chrysophana placida*: female, 8–11th, internal, ×220.



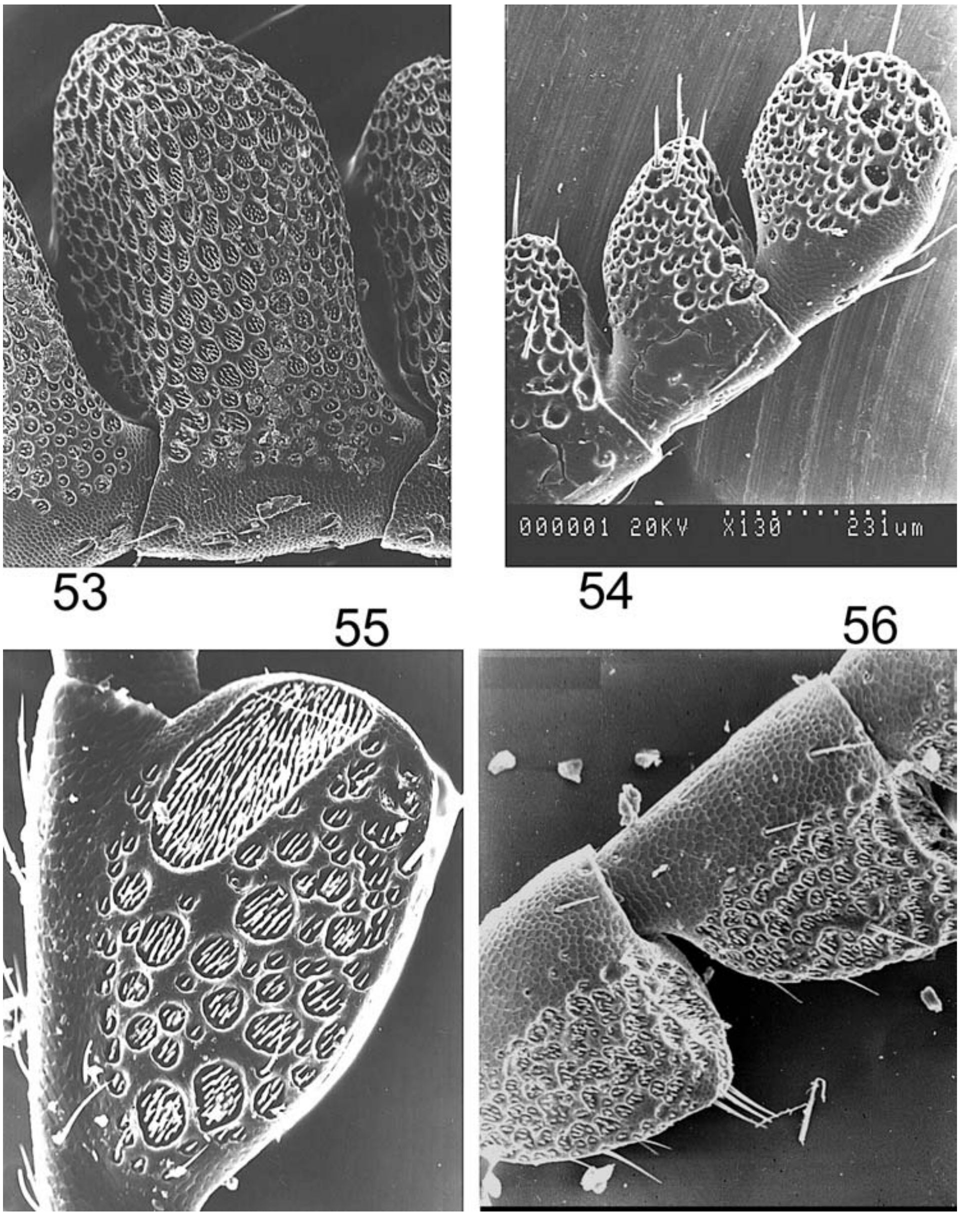
Figs 41–44: *Xyroscelis*, *Astraeus*, *Prospheres*, *Vadonaxia*. – 41. *Xyroscelis crocata*: 9–11th, internal, ×350; 42. *Astraeus flavopictus*: 7–8th, internal, ×200; 43. *Prospheres aurantiopictus*: 7th, internal, ×250; 44. *Vadonaxia peyrierasi*: 8–11th, internal, ×200.



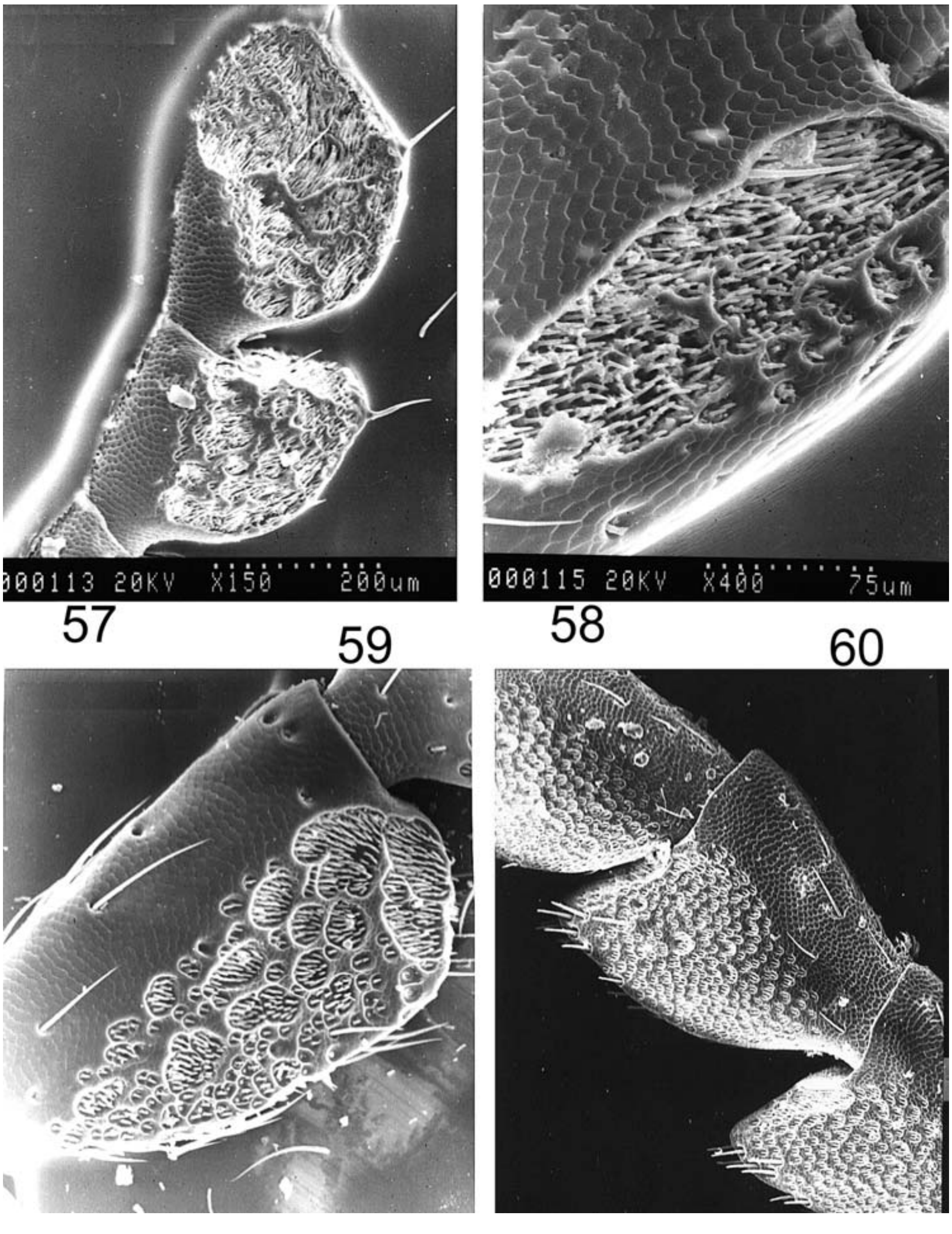
Figs 45–48: *Paratassa*, *Ampheremus*, *Nanularia*, *Chalcoplia*. –45. *Paratassa coraebiformis*: male, 8–11th, internal, ×170; 46. *Ampheremus cylindricollis*: 6–8th, internal, ×500; 47. *Nanularia brunneata*: male, 7–11th, internal, ×100; 48. *Chalcoplia auripilis*: 10th, internal, ×450.



Figs 49–52: *Descarpentriesiola*, *Embrikillium*, *Asamia*. – 49, 50. *Descarpentriesiola freyi*: male; 49: 7–11th, internal, ×130; 50: 8–11th, external, ×200; 51. *Embrikillium mirandum*: 9–11th, internal, ×130; 52. *Asamia insolita*: 6–8th, internal, ×100.



Figs 53–56: *Agelia*, *Micropistus*, *Iridotaenia*, *Sapaia*. – 53. *Agelia peteli*: 8th, internal, ×100; 54. *Micropistus microcephalus*: male, 9–11th, internal, ×130; 55. *Iridotaenia sulcata*: 5th, internal, ×200; 56. *Sapaia brodsky*: 7–8th, internal, ×150.



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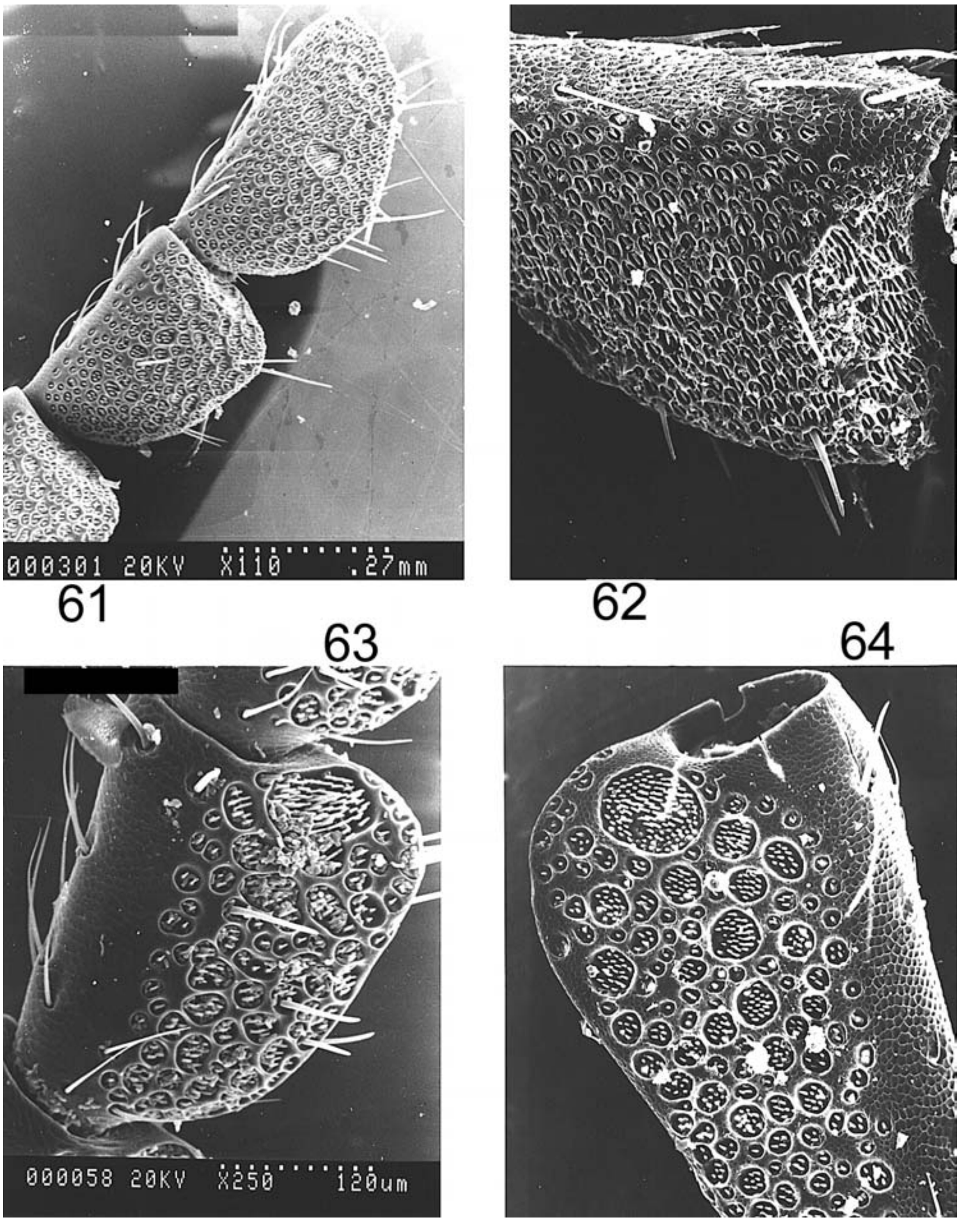
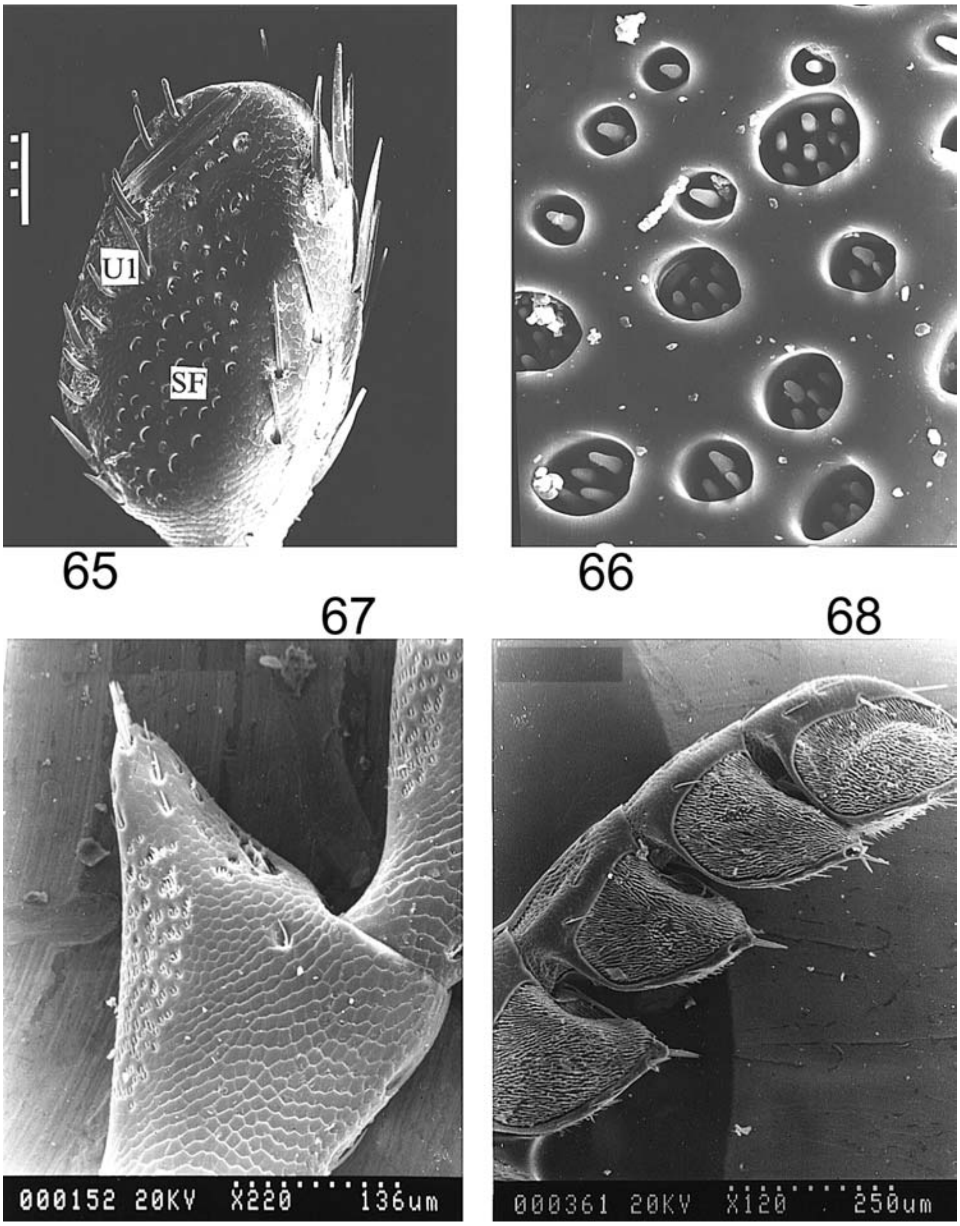
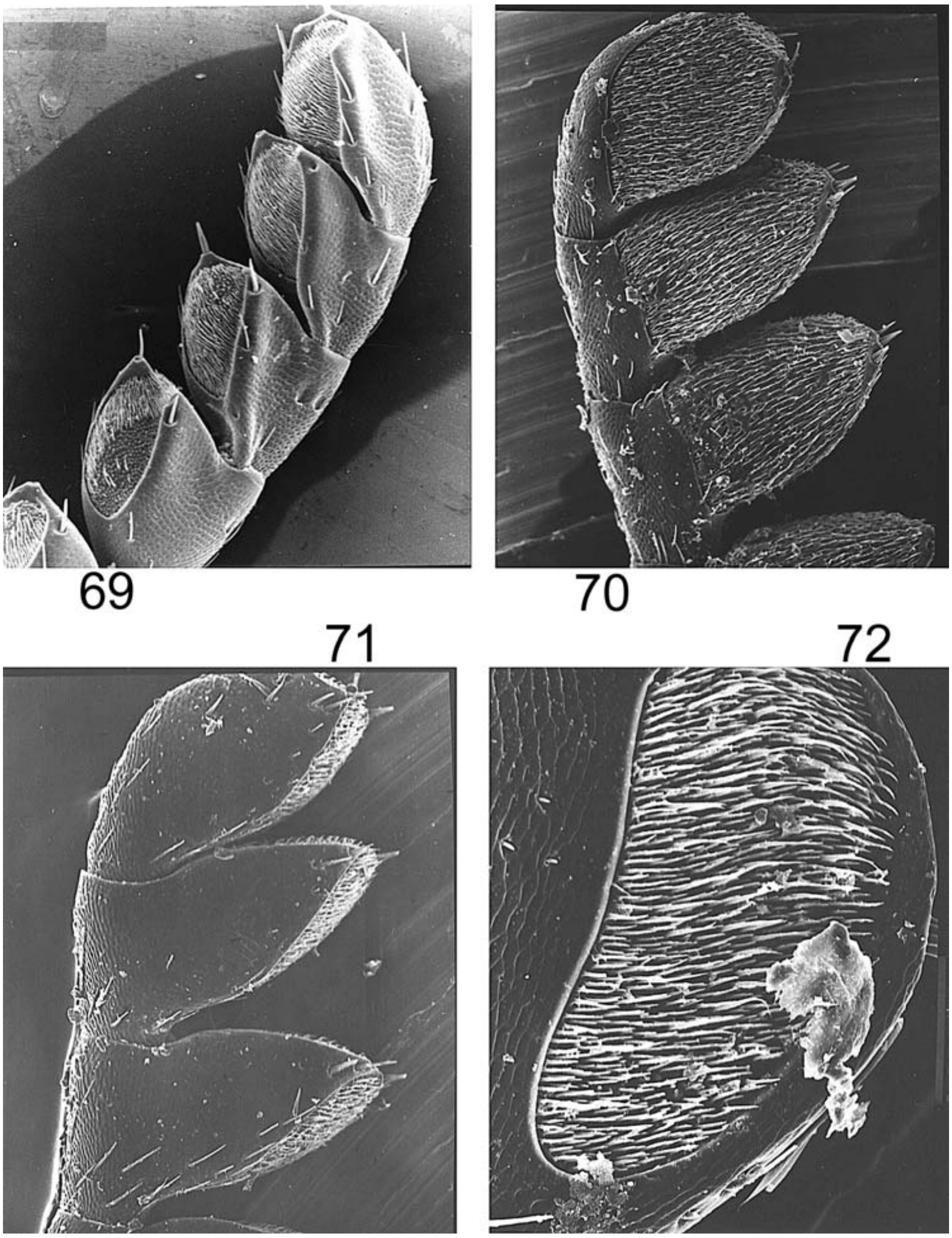


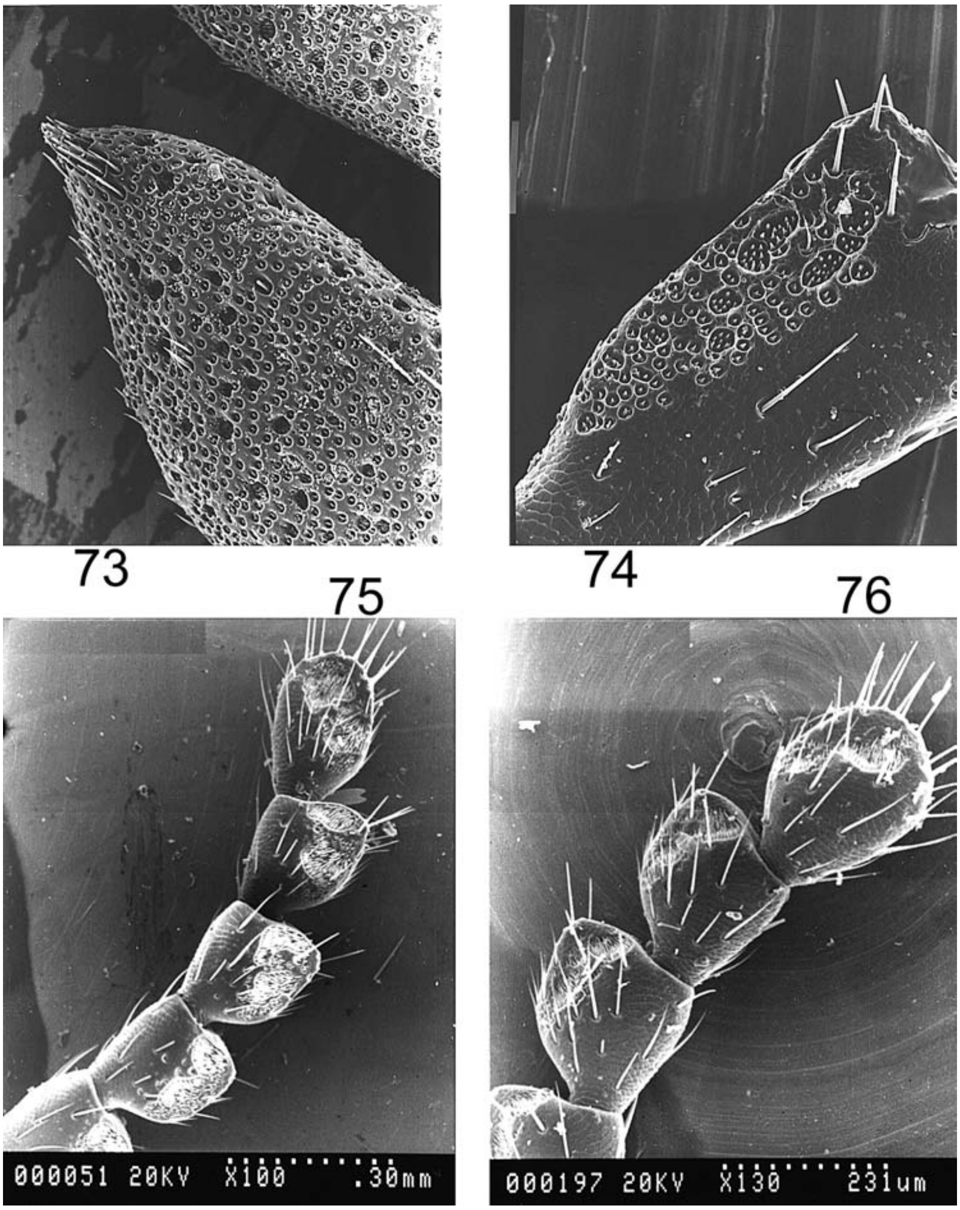
Figs 61–64: *Cyphogastrella*, *Nipponobuprestis*, *Tamamushia*, *Chalcophoropsis*. – 61. *Cyphogastrella gratiosissima*: 9–11th, internal, ×110; 62. *Nipponobuprestis amabilis*: 9th, internal, ×200; 63. *Tamamushia virida*: 10th, internal, ×250; 64. *Chalcophoropsis quadrifoveolata*: 5th, internal, ×150.



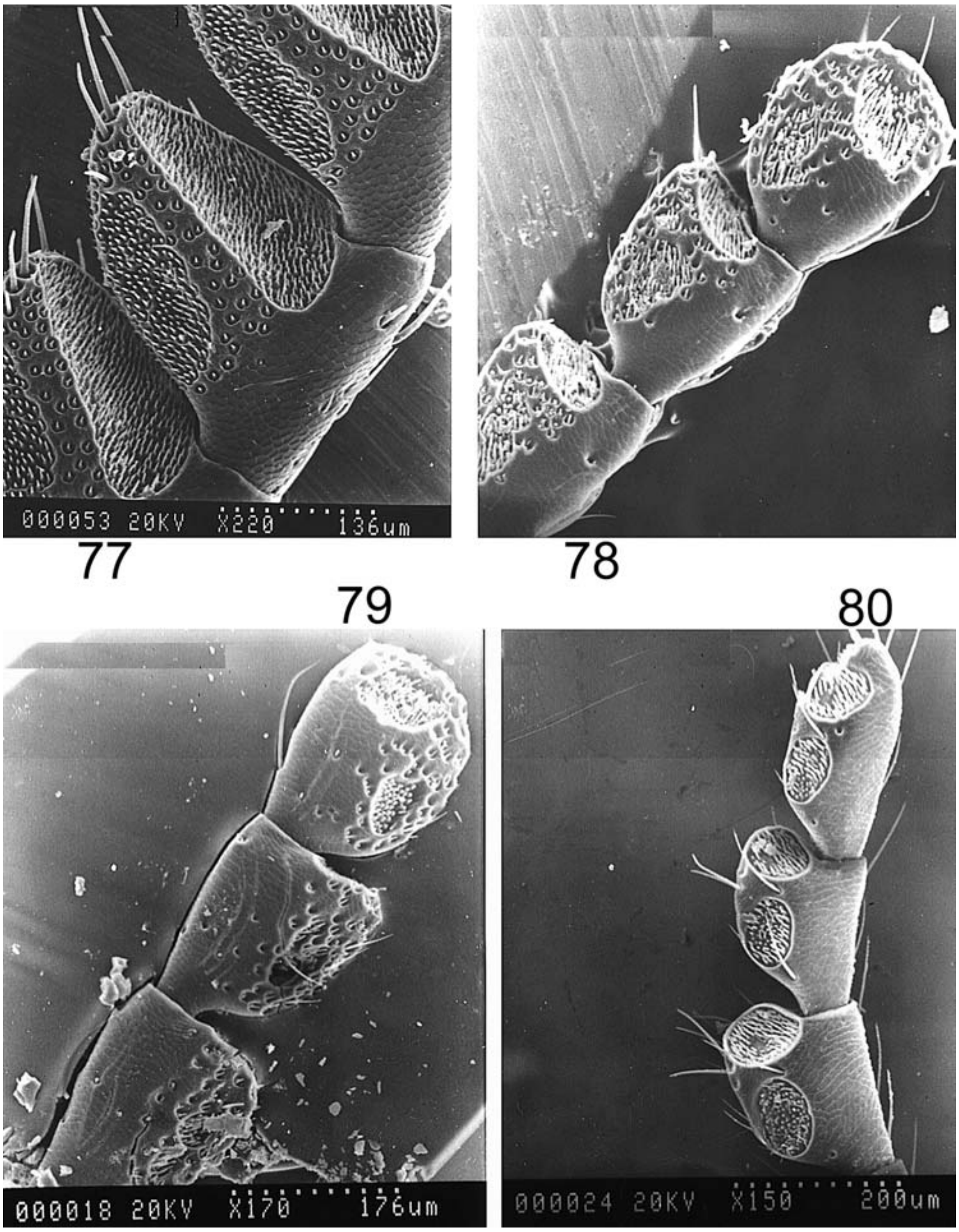
Figs 65–68: *Chalcophora*, *Chlorophorella*, *Evides*. – 65, 66. *Chalcophora mariana*, 65: male, 11th, internal, showing sensory field (SF) and zone of shortened uniporous (U1) sensillae, ×200; 66: female, 8th, internal, showing sensory field of little pits, ×1000; 67. *Chlorophorella gerlingi*: 6th, internal, ×220; 68. *Evides kraatzi*: 8–11th, internal, ×120.



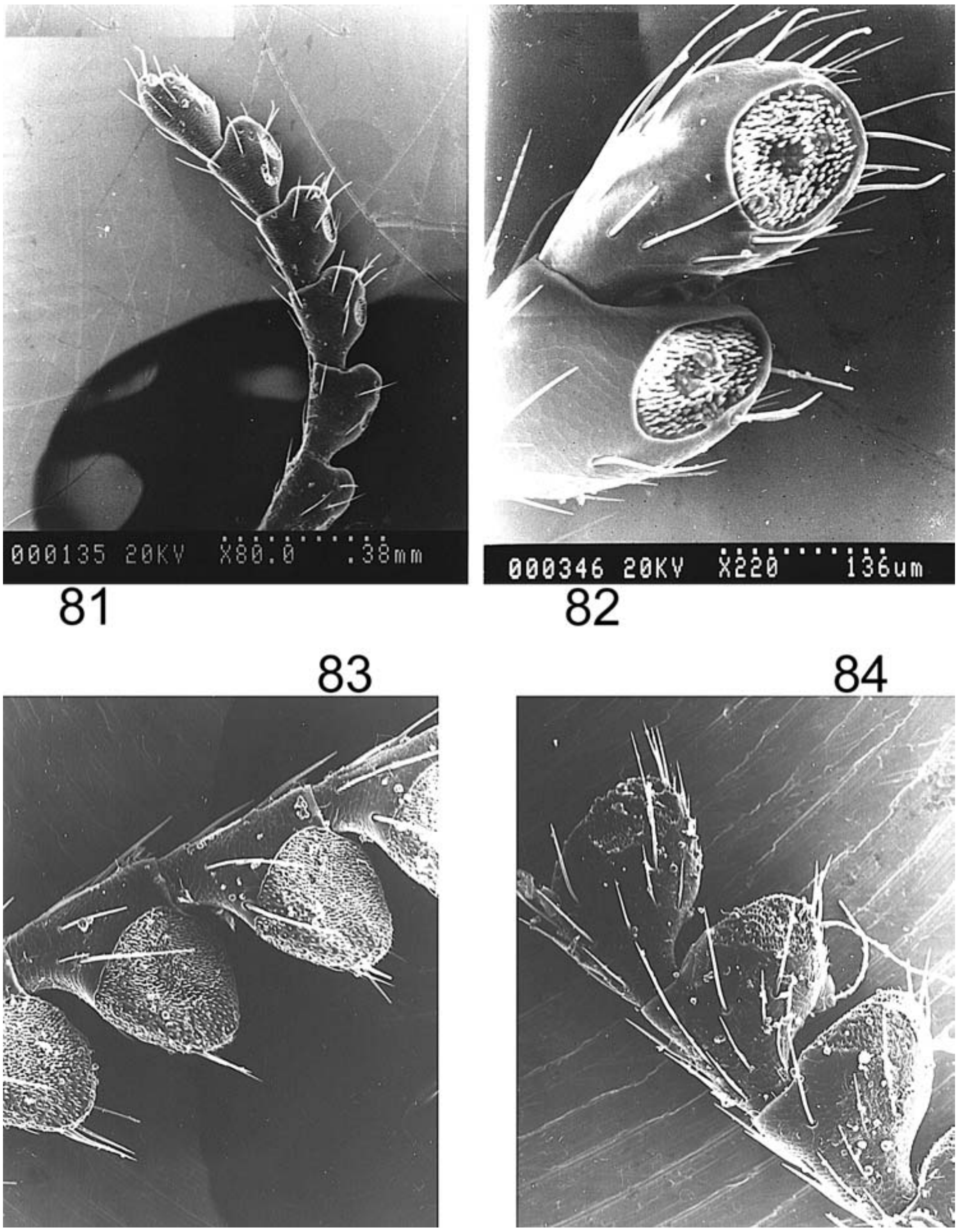
Figs 69–72: *Evides*. – 69. *Evides kraatzi*: 8–11th, external, ×110; 70–72. *E. gambiensis*, 70: 9–11th, internal, ×100; 71: 9–11th, external, ×100; 72: 4th, internal, ×300.



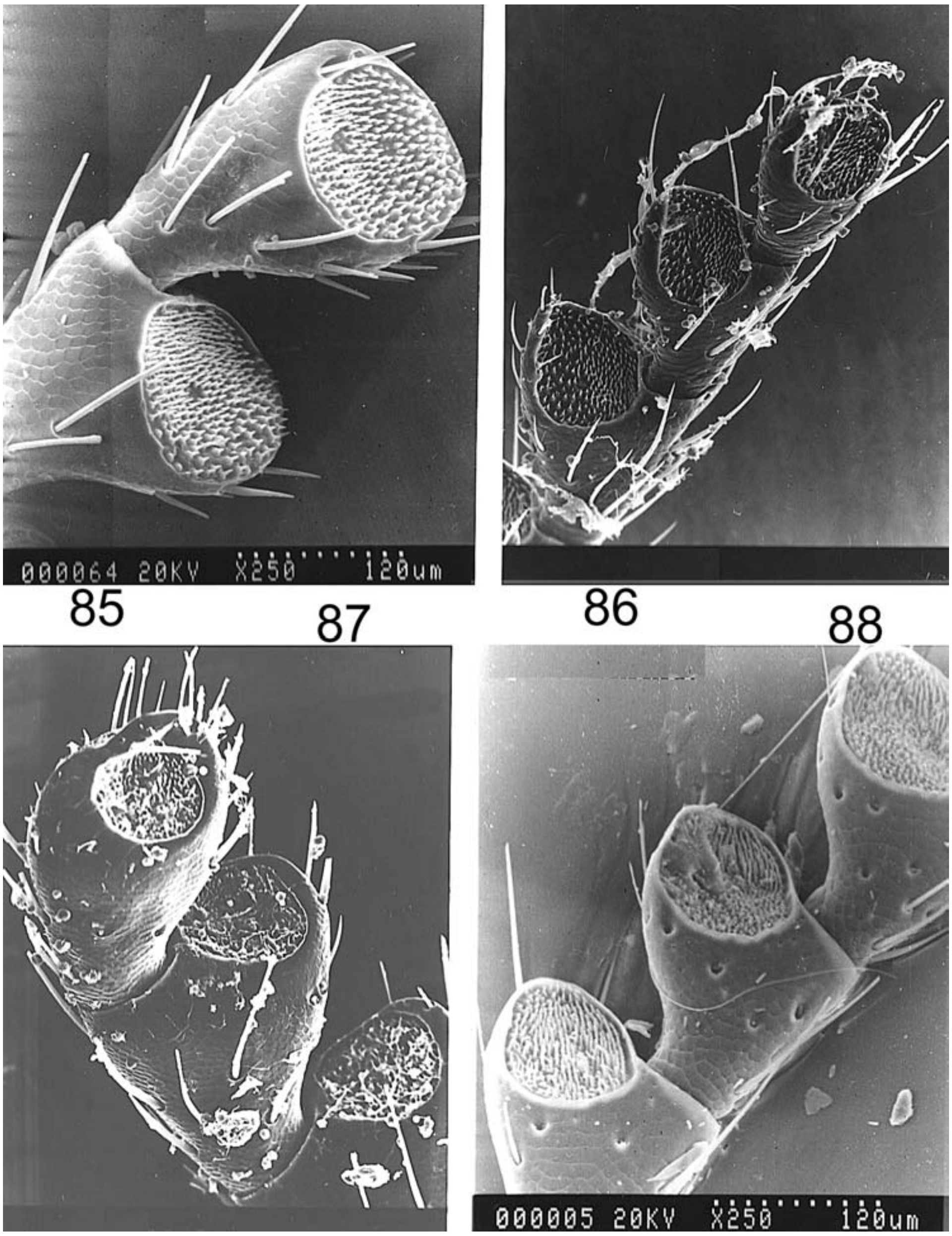
Figs 73–76: *Euchroma*, *Hypoprasis*, *Eupodalecia*. – 73. *Euchroma gigantea*: 9th, internal, ×100; 74. *Hypoprasis harpagon*: 8th, internal, ×200; 75, 76. *Eupodalecia minarum*, 75: 8–11th, internal, ×100; 76: 8–11th, external, ×130.



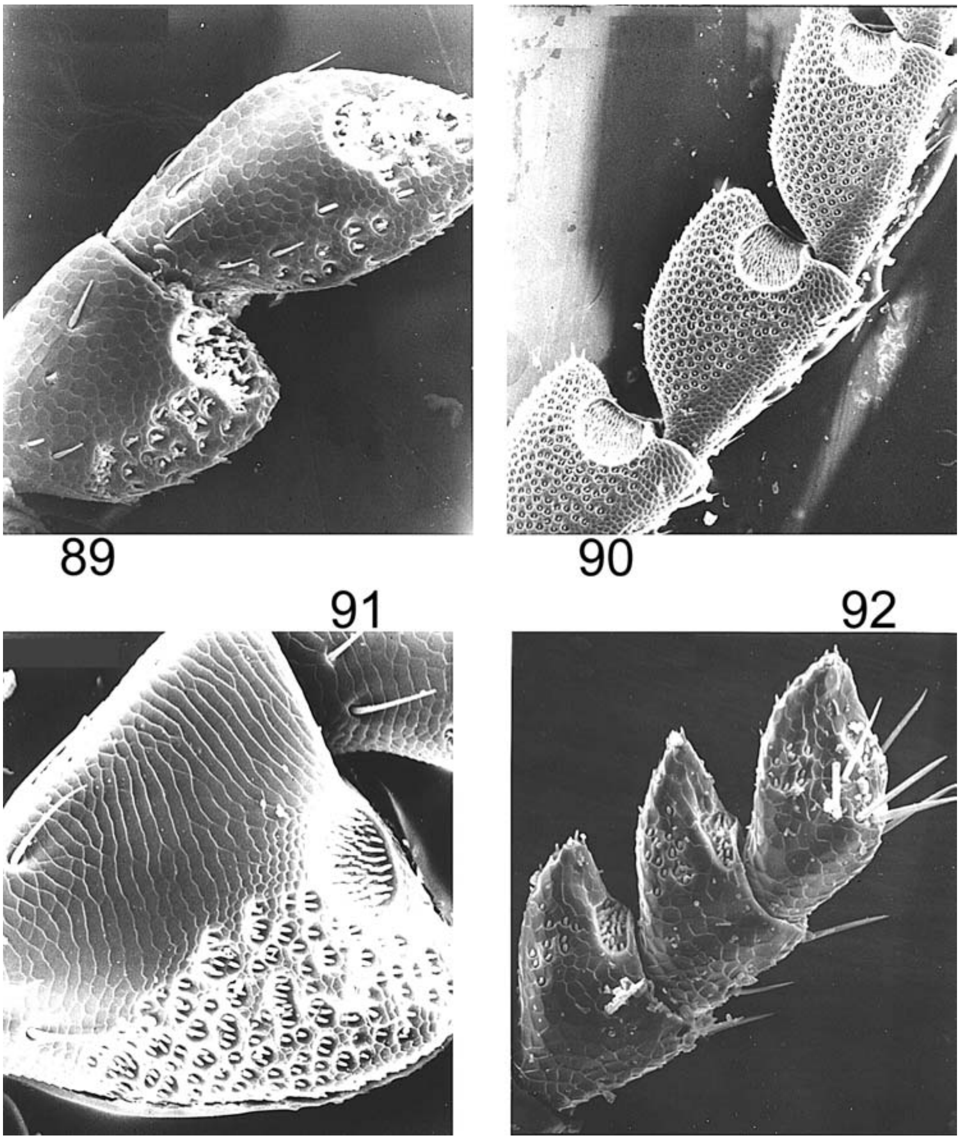
Figs 77–80: *Chrysesthes*, *Euplectalecia*, *Cinyra*. – 77. *Chrysesthes gymnopleura*: 7–9th, internal, ×220; 78, 79. *Euplectalecia pulverulenta*, 9–11th: 78: internal, ×170; 79: external, ×170; 80 *Cinyra obenbergeri*: 9–11th, internal, ×150.



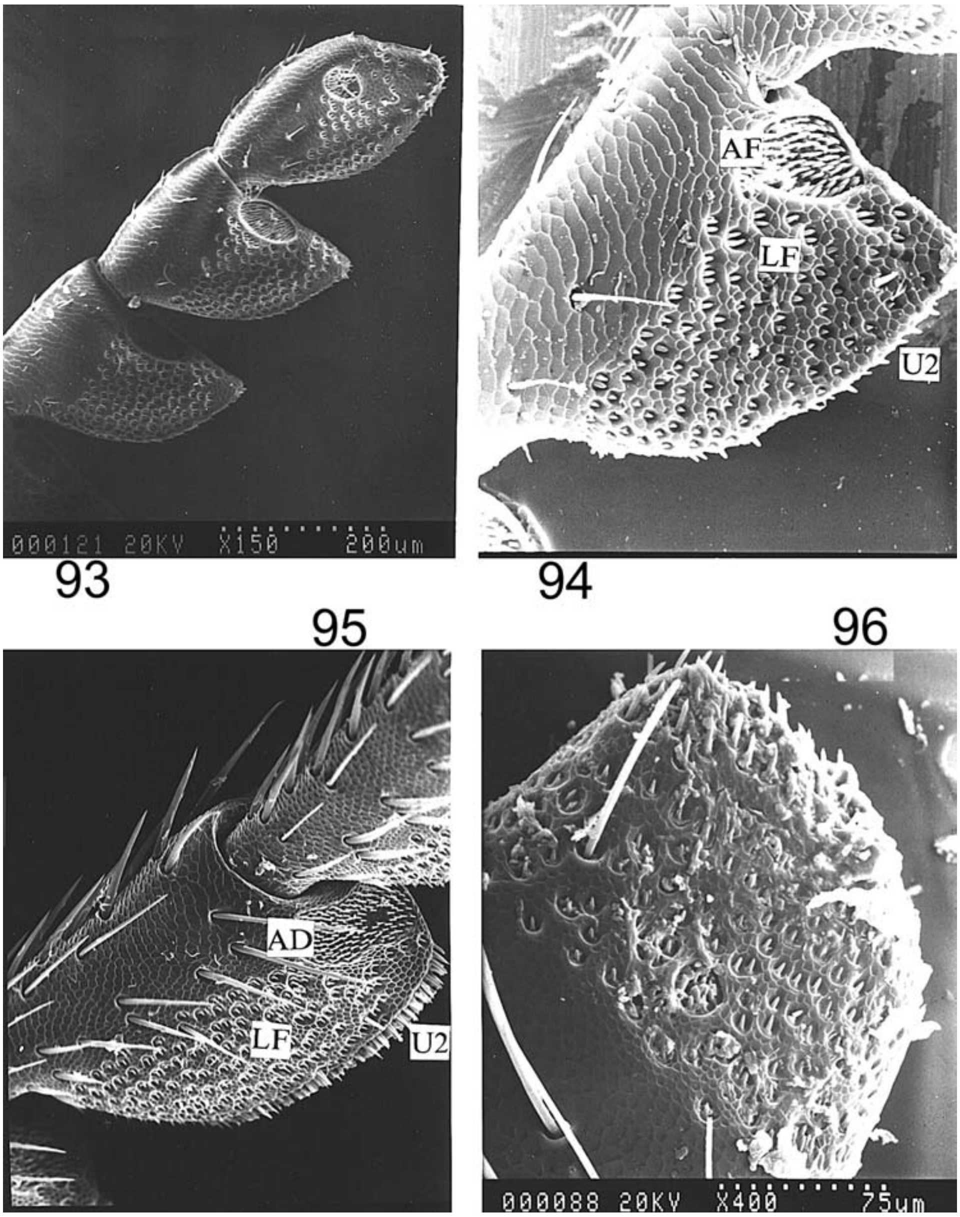
Figs 81–84: *Cinyra*, *Saundersina*, *Palmar*. – 81. *Cinyra obenbergeri*: 6–11th, external, ×80; 82. *Saundersina modesta*: 10–11th, internal, ×220; 83, 84. *Palmar* (*Scintillatrix*) *limbata*: female; 83: 7–9th, internal, ×100; 84: 9–11th, external, ×100.



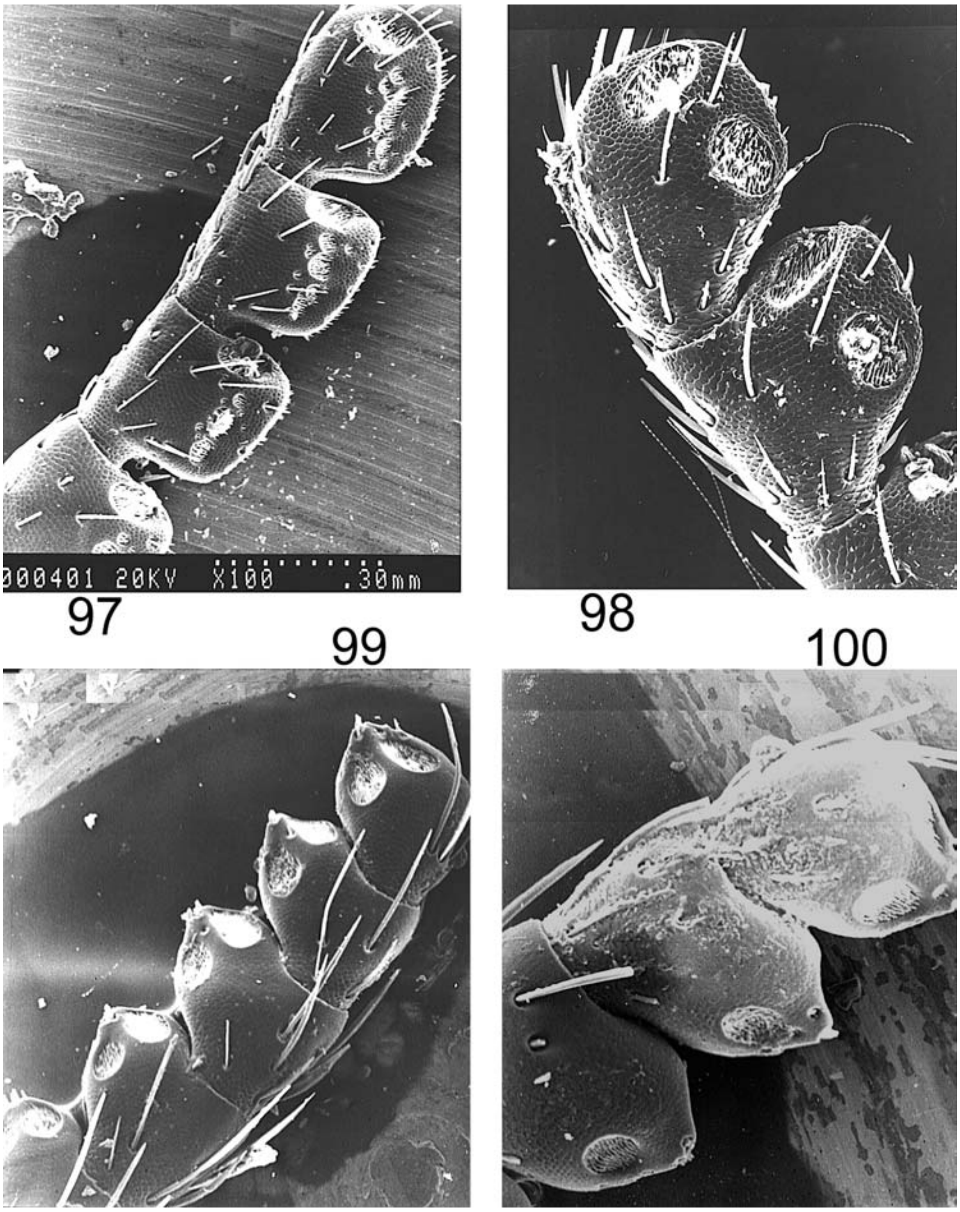
Figs 85–88: *Palmar*, *Poecilonota*, *Nesotrinchus*. – 85. *Palmar* (*Scintillatrix*) *chinganensis*: female, 10–11th, internal, ×250; 86. *P.* (s.str.) *virgata*: 9–11th, internal, ×150; 87. *Poecilonota variolosa*: 9–11th, internal, ×150; 88. *Nesotrinchus orientalis*: 7–9th, internal, ×250.



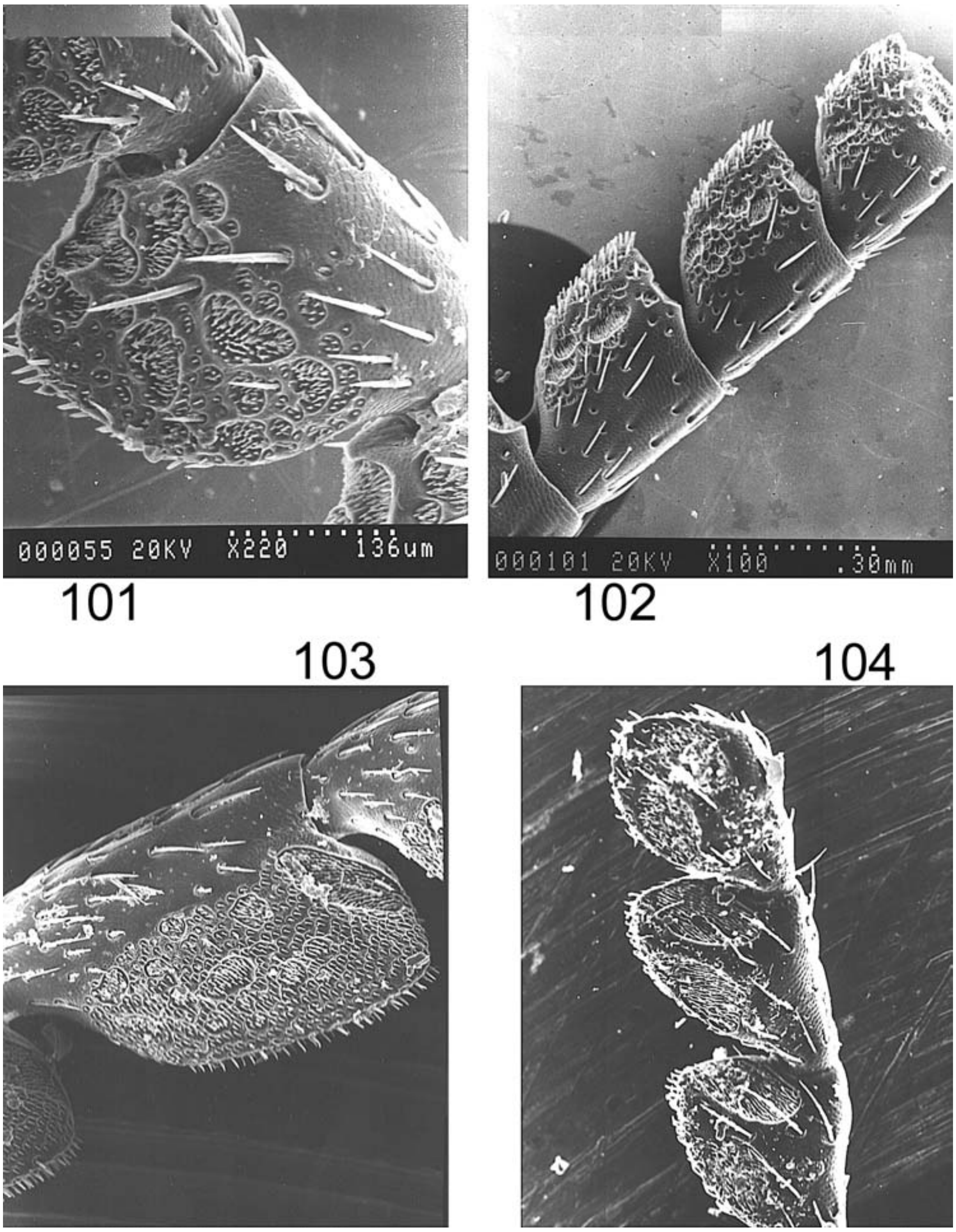
Figs 89–92: *Genestia*, *Armenosoma*, *Evagora*, *Sphenoptera*. – 89. *Genestia semenovi*, 10–11th, internal, ×350; 90. *Armenosoma atrum*: 6–8th, internal, ×170; 91. *Evagora amorpha*: 8th, internal, ×400; 92. *Sphenoptera* (*Sphenopterella*) *margaritae*: 9–11th, internal, ×350.



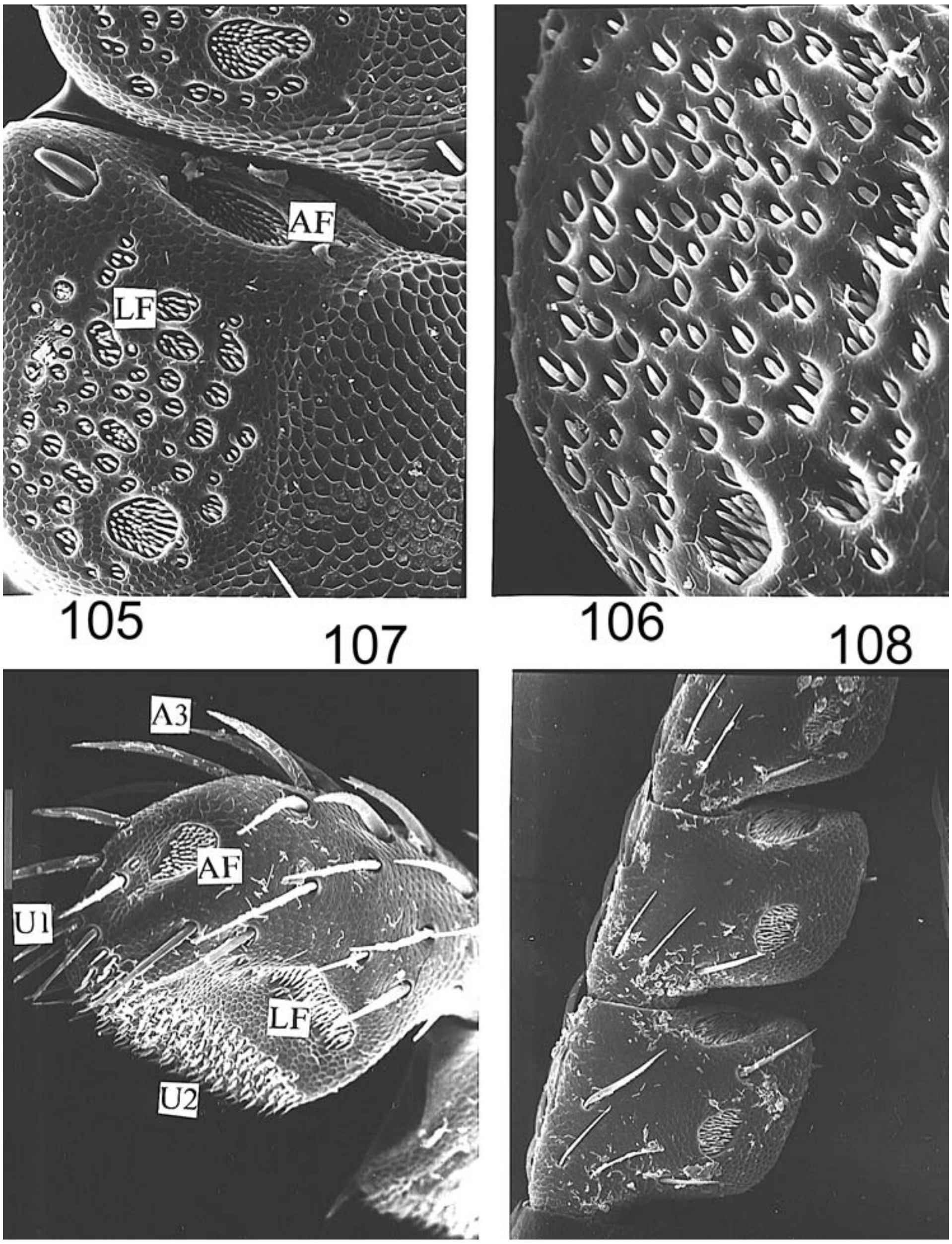
Figs 93–96: *Sphenoptera*, *Hippomelas*, *Achardella*. – 93. *S.* (s.str.) *glabrata*: 9–11th, internal, ×150; 94. *S.* (*Chrysoblemma*) *beckeri*: 8th, internal, showing apical fossa (AF), lateral field (LF) and short uniporous sensillae (U2), ×400; 95. *Hippomelas* (s.str.) *planicauda*: 10th, internal, showing apical depression (AD), lateral field (LF), and zone of short uniporous sensillae (U2), ×150; 96. *Achardella americana*: 8th, external, ×400.



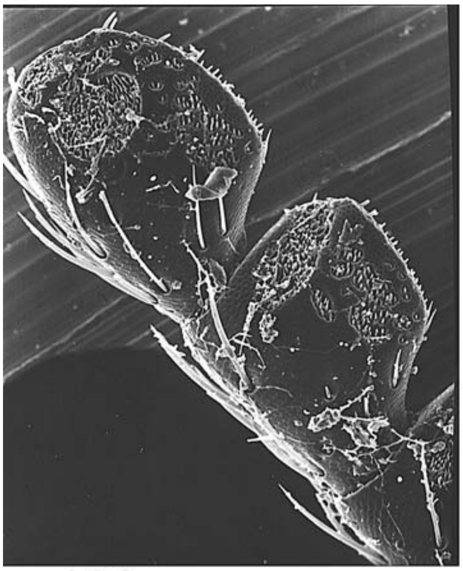
Figs 97–100: *Pseudolampetis*, *Ectinogonia*, *Chalcopoecila*. – 97. *Pseudolampetis circumsulcata*: 8–11th, internal, ×100; 98. *Ectinogonia buqueti speciosa*: 10–11th, internal, ×150; 99, 100. *Chalcopoecila ornata*, 99: 7–11th, internal, ×100; 100: 9–11th, external, ×150.



Figs 101–104: *Asidoptera*, *Pseudoperotis*, *Polybothris*. – 101. *Asidoptera monstruosa*: 10th, internal, ×220; 102. *Pseudoperotis* sp.: 9–11th, external, ×100; 103. *Polybothris sulcicollis*: 8th, internal, ×100; 104. *P. staudingeri*: 9–11th, internal, showing apical and lateral fossae, ×100.



Figs 105–108: *Capnodis*, *Cyphosoma*, *Perotis*, *Lampetis*. – 105. *Capnodis tenebrionis*: 8–9th, internal, showing apical fossa (AF) and lateral field (LF), ×300; 106. *Cyphosoma tataricum*: 7th, internal, showing lateral field, ×700; 107. *Perotis lugubris*: 11th, external, showing apical (AF) and lateral (LF) fossae, zone of short (U2) and normal uniporous (U1), and aporous (A3) sensillae, ×200; 108. *Lampetis argentata*: 9–11th, internal, ×150.



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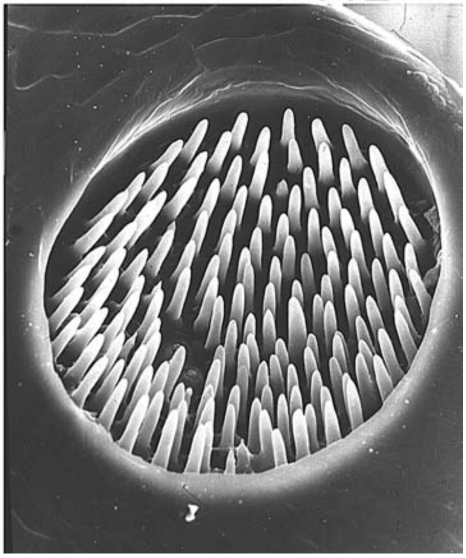

Figs 109–112: *Latipalpis*, *Dicerca*. – 109. *Latipalpis* (s.str.) *plana*: 10–11th, internal, showing subapical fossae and lateral fields, ×150; 110. *L.* (*Palpilatis*) *plasoni*: 5–6th, internal, showing the formation of subapical fossae, ×200; 111, 112. *Dicerca* (s.str.) *furcata*, 111: 8–11th, internal, ×100; 112: 9th, internal, showing subapical fossa, ×1000.

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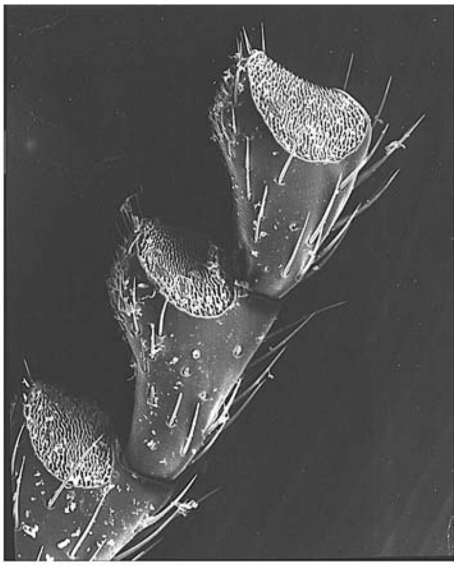

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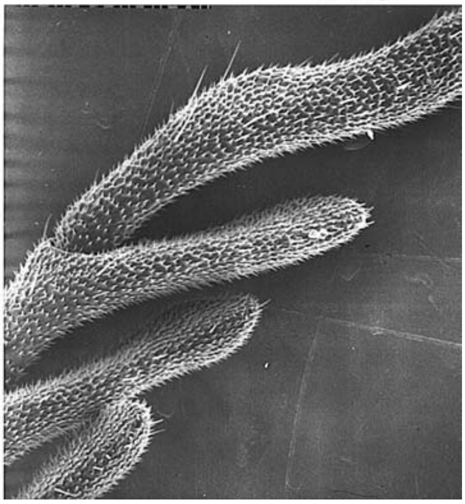
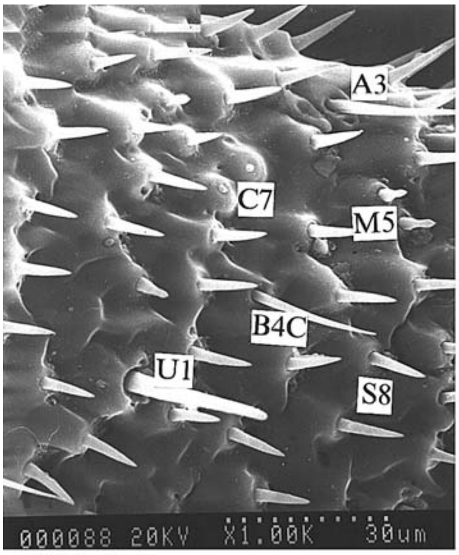


Figs 113–116: *Dicercomorpha*, *Cardiaspis*, *Haplotrinchus*, *Mendizabalia*. – 113. *Dicercomorpha albosparsa*: 9–11th, internal, ×100; 114. *Cardiaspis mouhoti*: 9–11th, internal, ×100; 115. *Haplotrinchus inaequalis*: 8–11th, internal, ×110; – 116. *Mendizabalia germaini*: male, 8–11th, internal, ×110.



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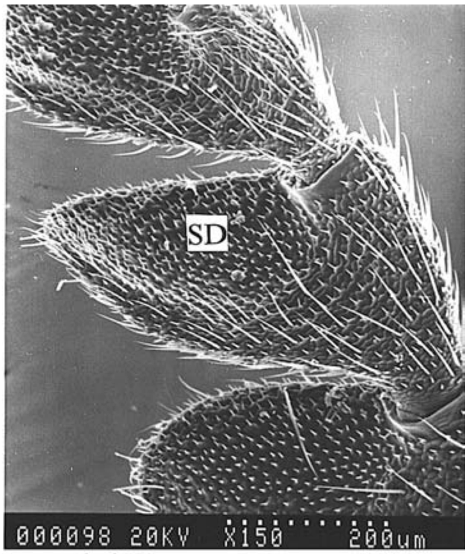

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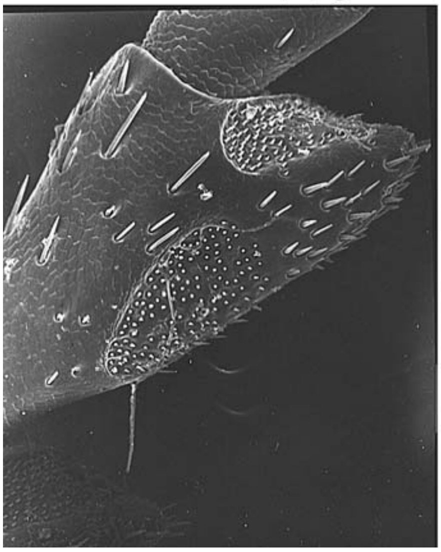

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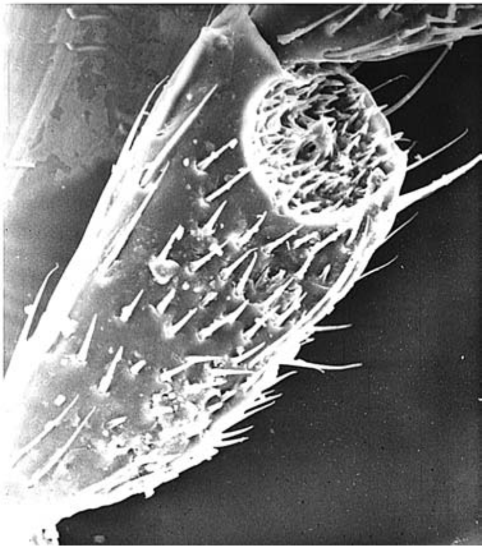
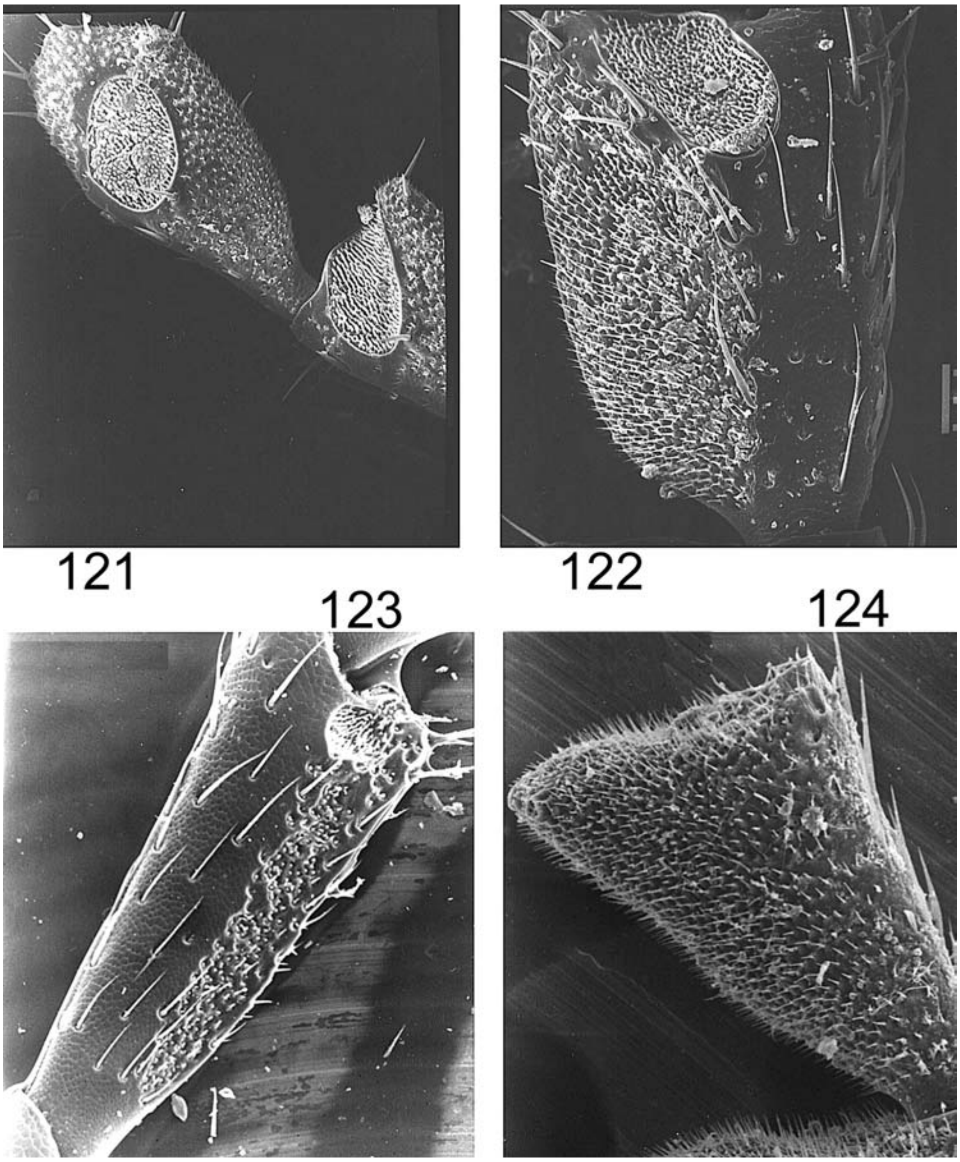
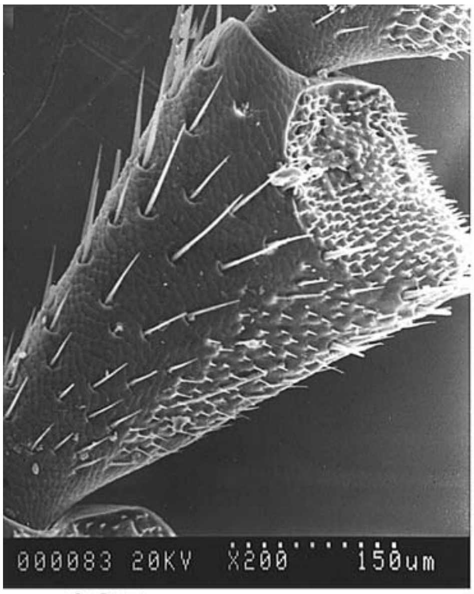


Figs 117–120: *Mendizabalia*, *Philandia*, *Julodimorpha*, *Pygicera*. – 117. *Mendizabalia germaini*: male, 11th, internal, showing basiconic (B4c, S8), multiporous (M5), campaniform (C7), uniporous (U1), and aporous (A3) sensillae at the apical part, ×1000; 118. *Philandia valdiviana*: male, 7–9th, internal, showing subapical depression (SD), ×150; 119. *Julodimorpha bakewelli*: 10th, internal, ×150; 120. *Pygicera scripta*: 10th, internal, ×350.



Figs 121–124: *Diadoxus*, *Cyria*, *Xenocyria*, *Neobuprestis*. – 121. *Diadoxus erythrurus*: 10–11th, internal, ×150; 122. *Cyria imperialis*: 9th, internal, ×150; 123. *Xenocyria vittigera*: 7th, internal, ×130; 124. *Neobuprestis australis*: male, 9th, internal, ×200.



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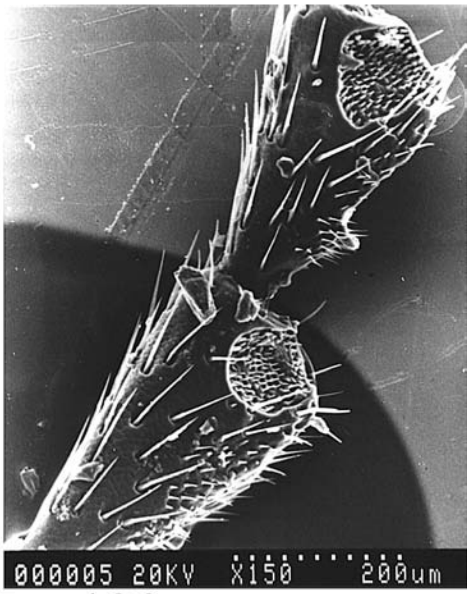

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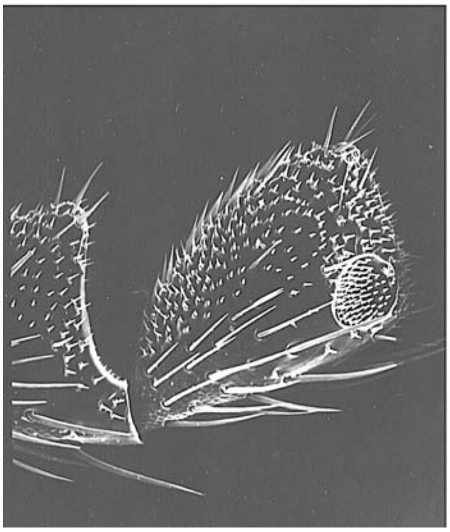

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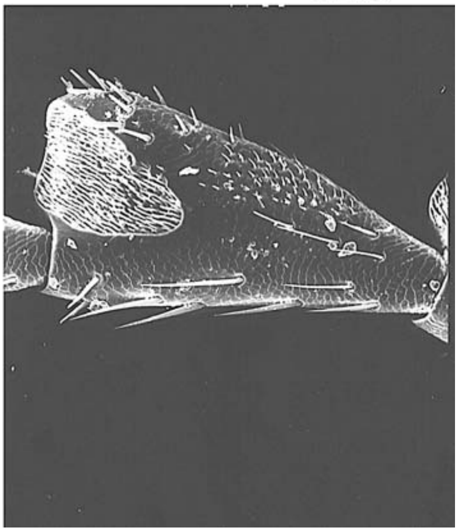
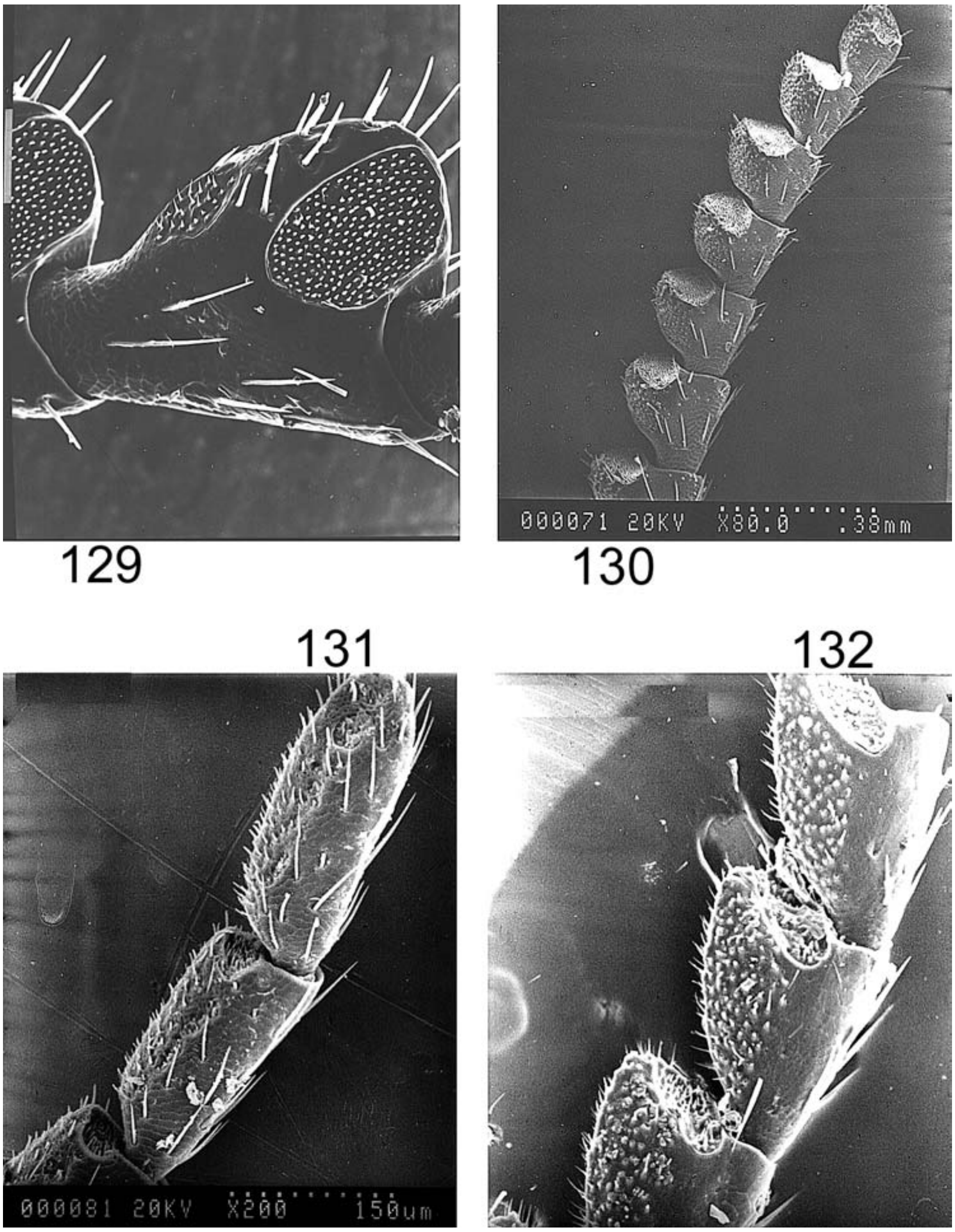
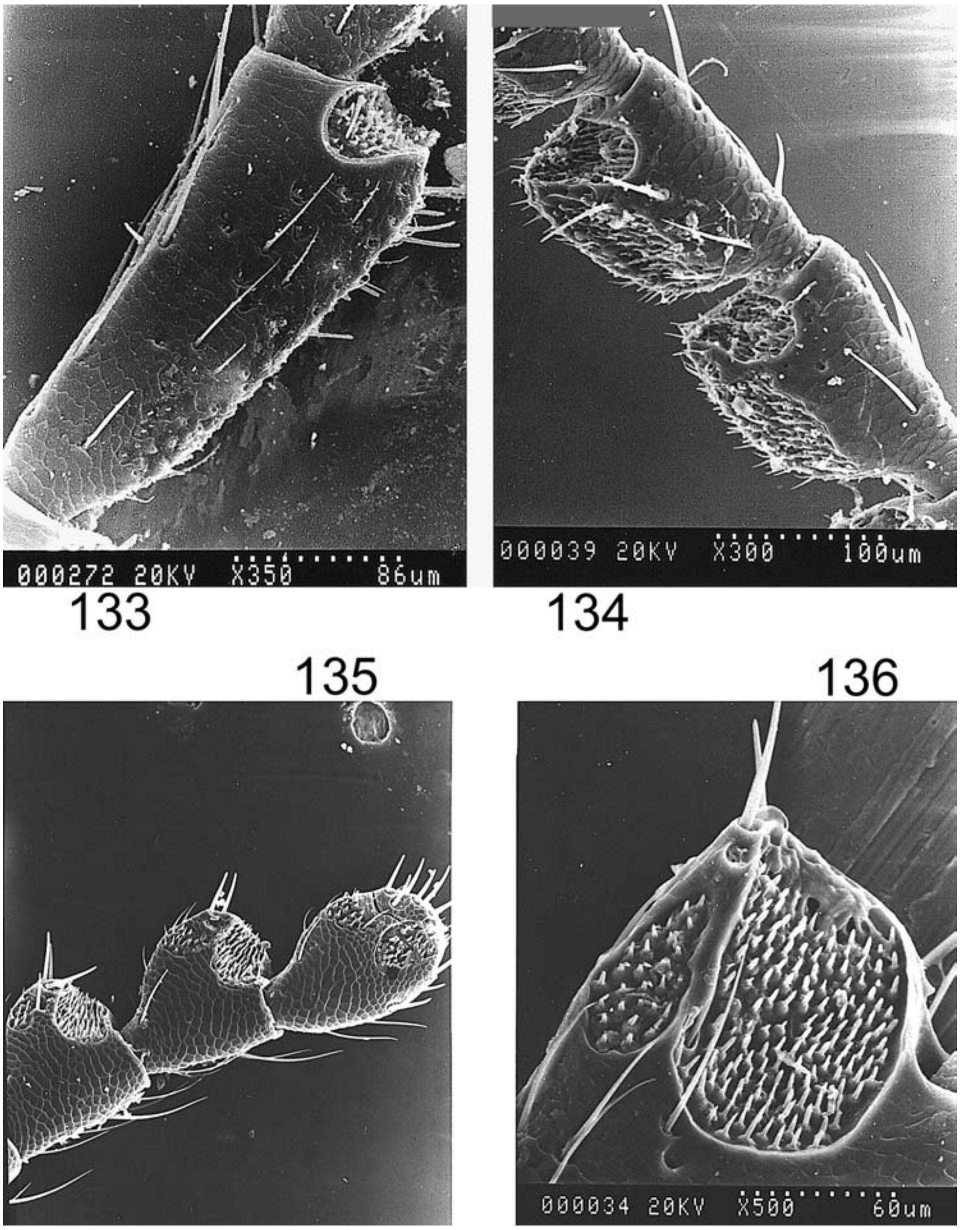


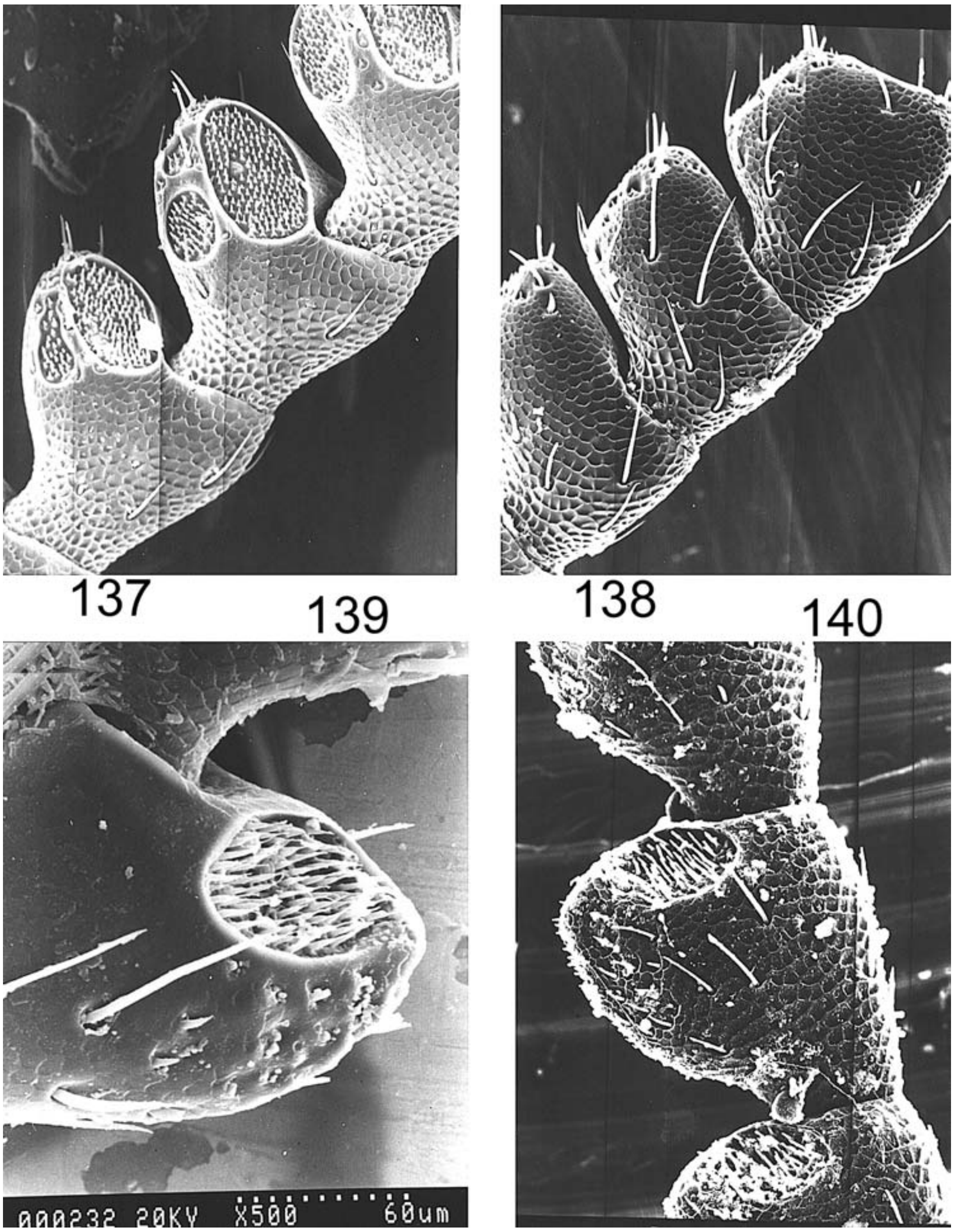
Figs 125–128: *Neobuprestis*, *Balthasarella*, *Trachykele*, *Buprestis*. – 125. *Neobuprestis australis*: female, 8th, internal, ×200; 126. *Balthasarella melandryoides*: female, 7–8th, internal, ×150; 127. *Trachykele blondeli*: 10–11th, external, ×150; 128. *Buprestis* (*Orthocheira*) *salomoni*: 10th, internal, ×150.



Figs 129–132: *Eurythyrea*, *Yamina*, *Lamprocheila*, *Phrixia*. – 129. *Eurythyrea aurata*: 5–6th, internal, ×200; 130. *Yamina sanguinea*: 5–11th, internal, ×80; 131. *Lamprocheila maillei*: 8–10th, internal, ×170; 132. *Phrixia* sp.: male, 9–11th, internal, ×200.



Figs 133–136: *Paraphrixia*, *Exagistus*, *Mixochlorus*, *Peronaemis*. – 133. *Paraphrixia purpurea*: 10th, internal, ×350; 134. *Exagistus igneiceps*: 7–8th, internal, ×300; 135. *Mixochlorus lateralis*: 9–11th, internal, ×200; 136. *Peronaemis monticola*: 5th, internal, ×500.



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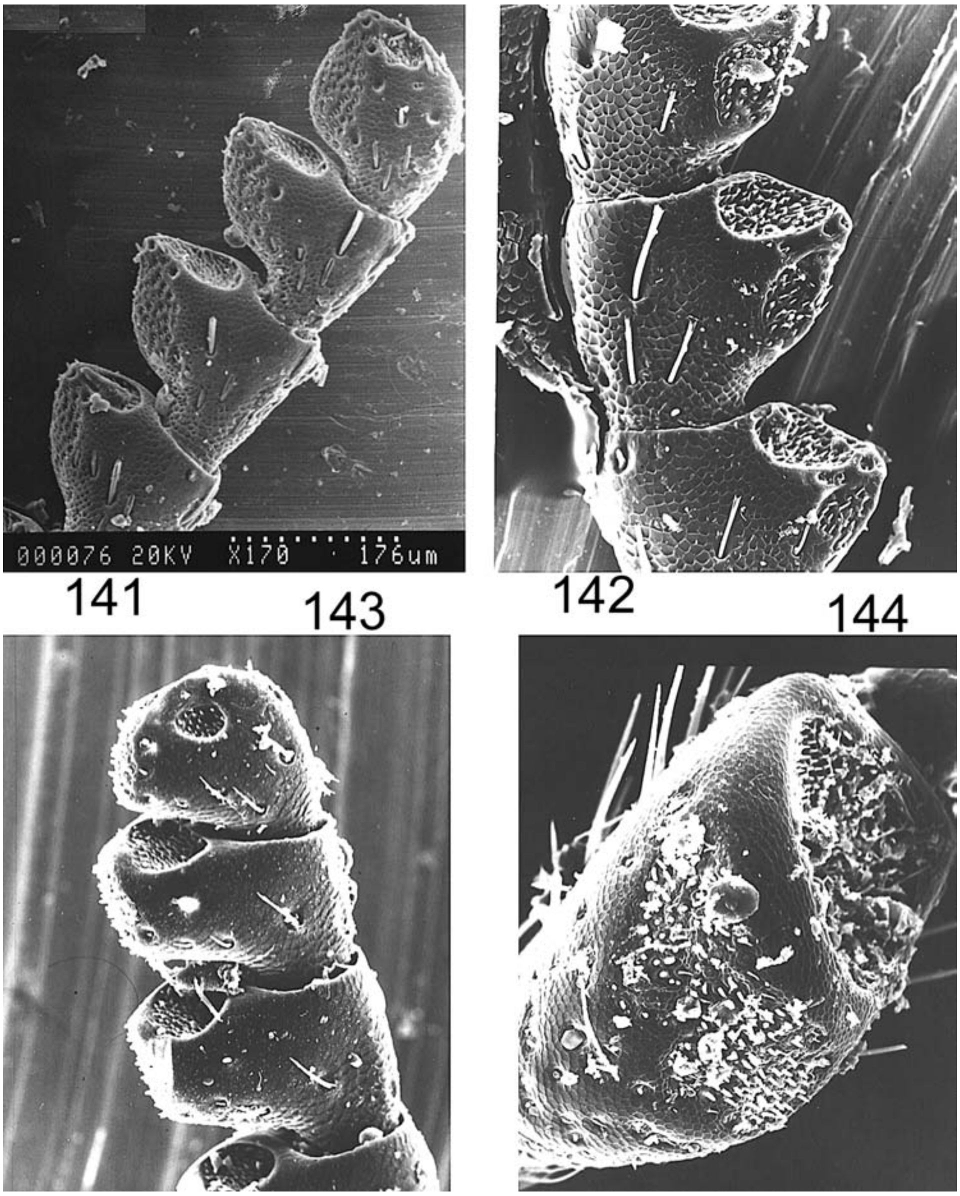
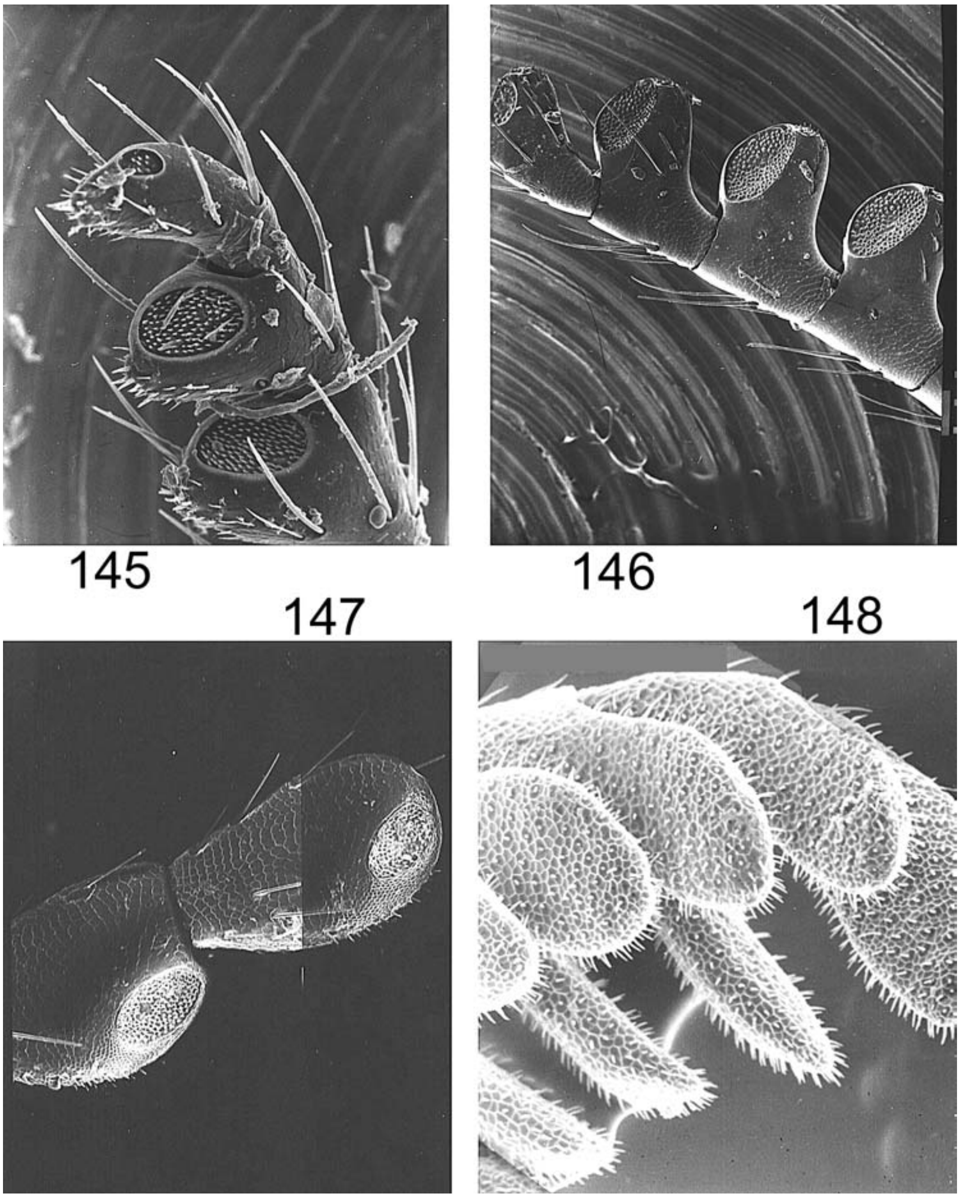
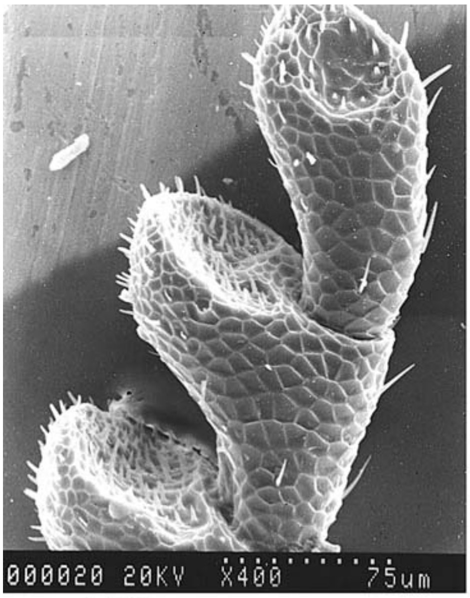


Figs 141–144: *Bubastes*, *Microcastalia*, *Calodema*. – 141. *Bubastes inconsistans*: 8–11th, internal, ×170; 142. *Bubastes iridescens*: 6–8th, internal, ×200; 143. *Microcastalia globithorax*: 8–11th, internal, ×200; 144. *Calodema regalis*: 9th, inner margin, ×200.



Figs 145–148: *Dactylozodes*, *Hiperantha*, *Conognatha*, *Hypostigmodera*. – 145. *Dactylozodes confus*: 9–11th, internal, ×200; 146. *Hiperantha* (s.str.) *haemorrhoa*: 8–11th, internal, ×1000; 147. *Conognatha pretiosissima*: 10–11th, internal, ×150; 148. *Hypostigimodera variegata*: male, 8–12th, internal, ×250.



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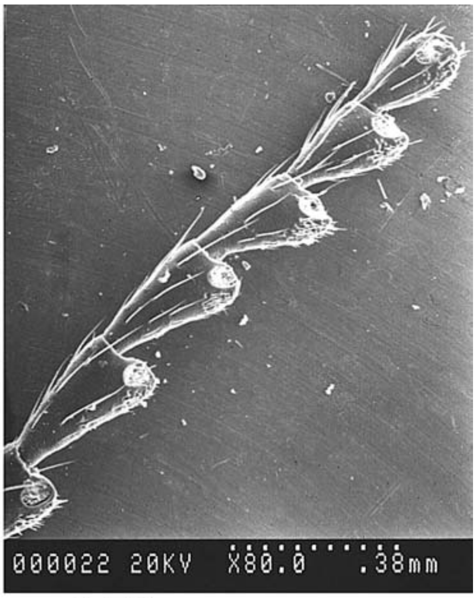

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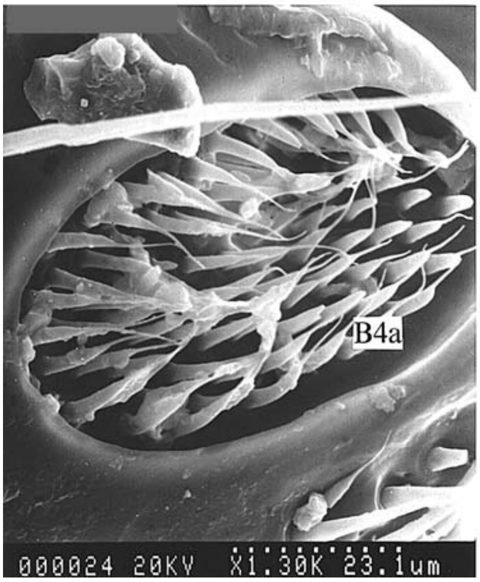

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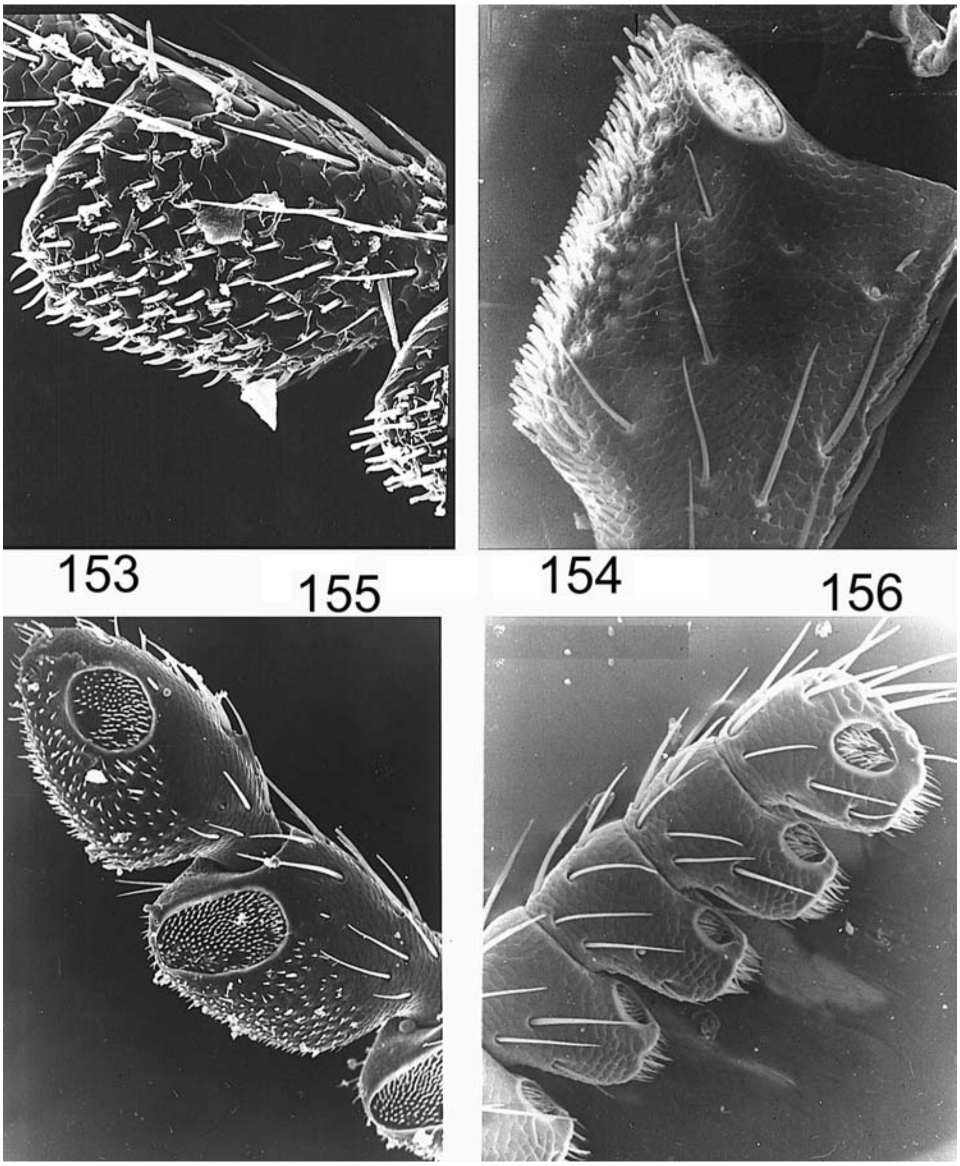
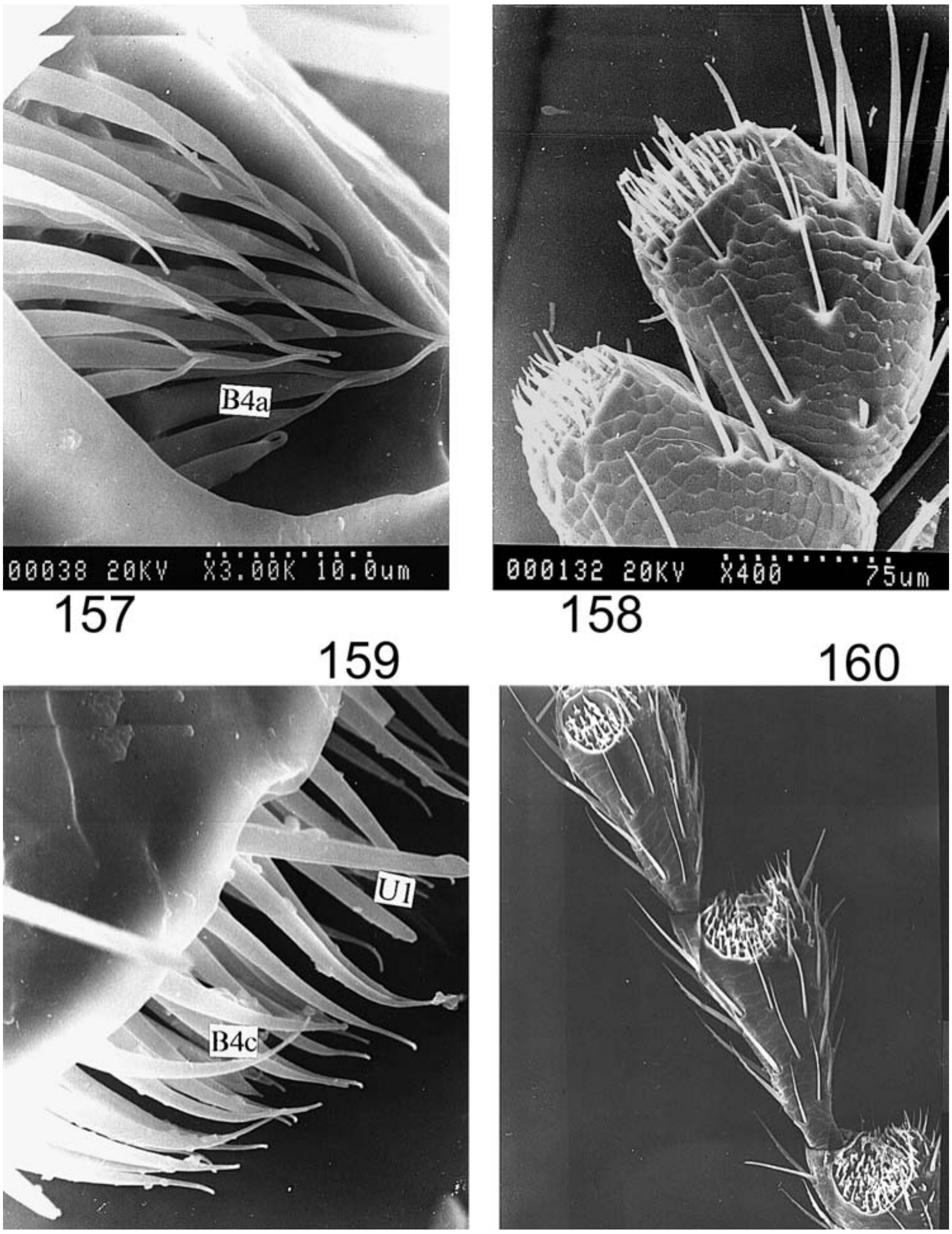
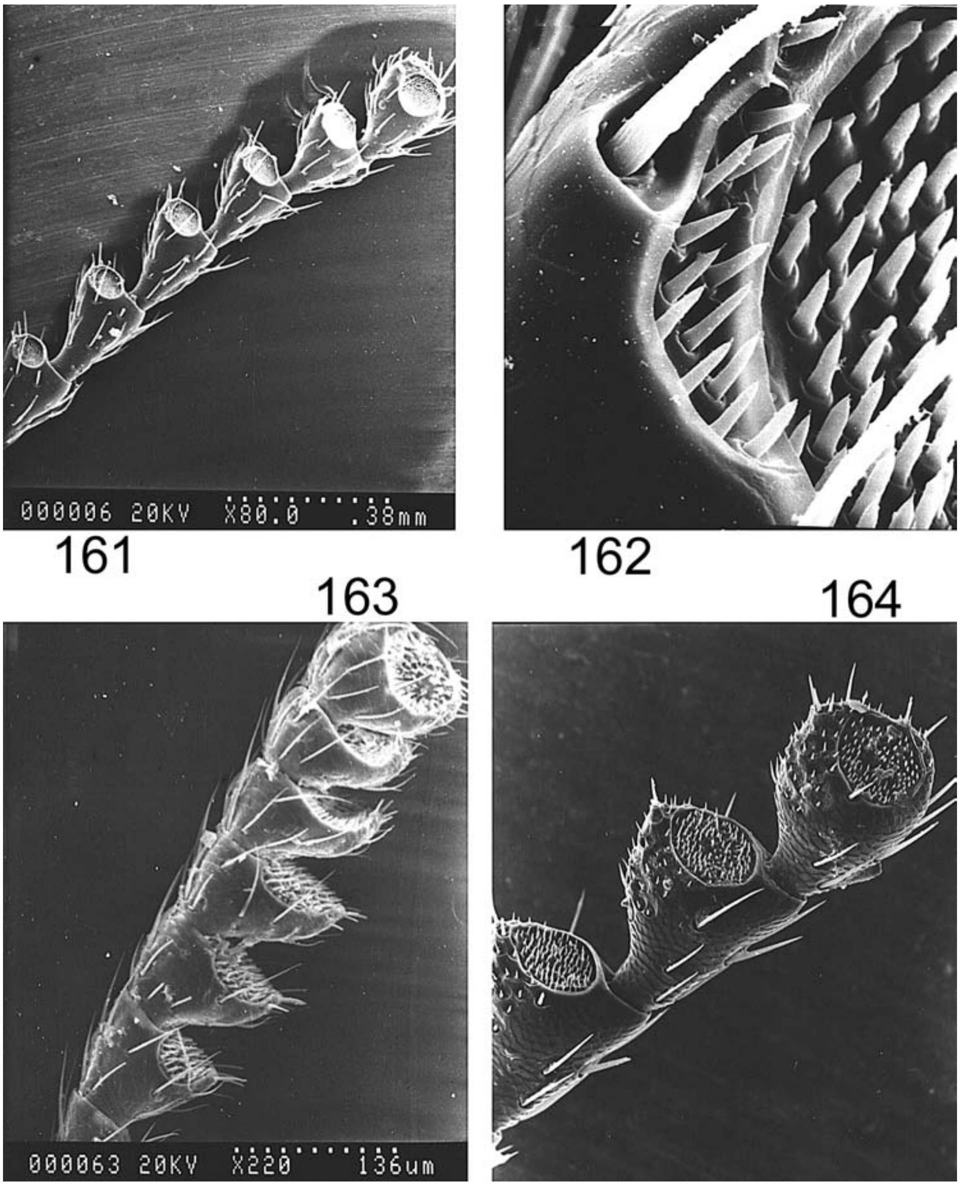
Figs 149–152: *Hypostigmodera*, *Aglaostola*, *Spectralia*. – 149. *Hypostigimodera variegata*: female, 10–12th, internal, ×400; 150, 151. *Aglaostola tereticollis*: 150: 6–11th, internal, ×80; 151: 6th, internal, showing apical fossa comprising modified basiconic B4a sensillae, ×1300; 152. *Spectralia purpurascens*: 10–11th, internal, ×450.



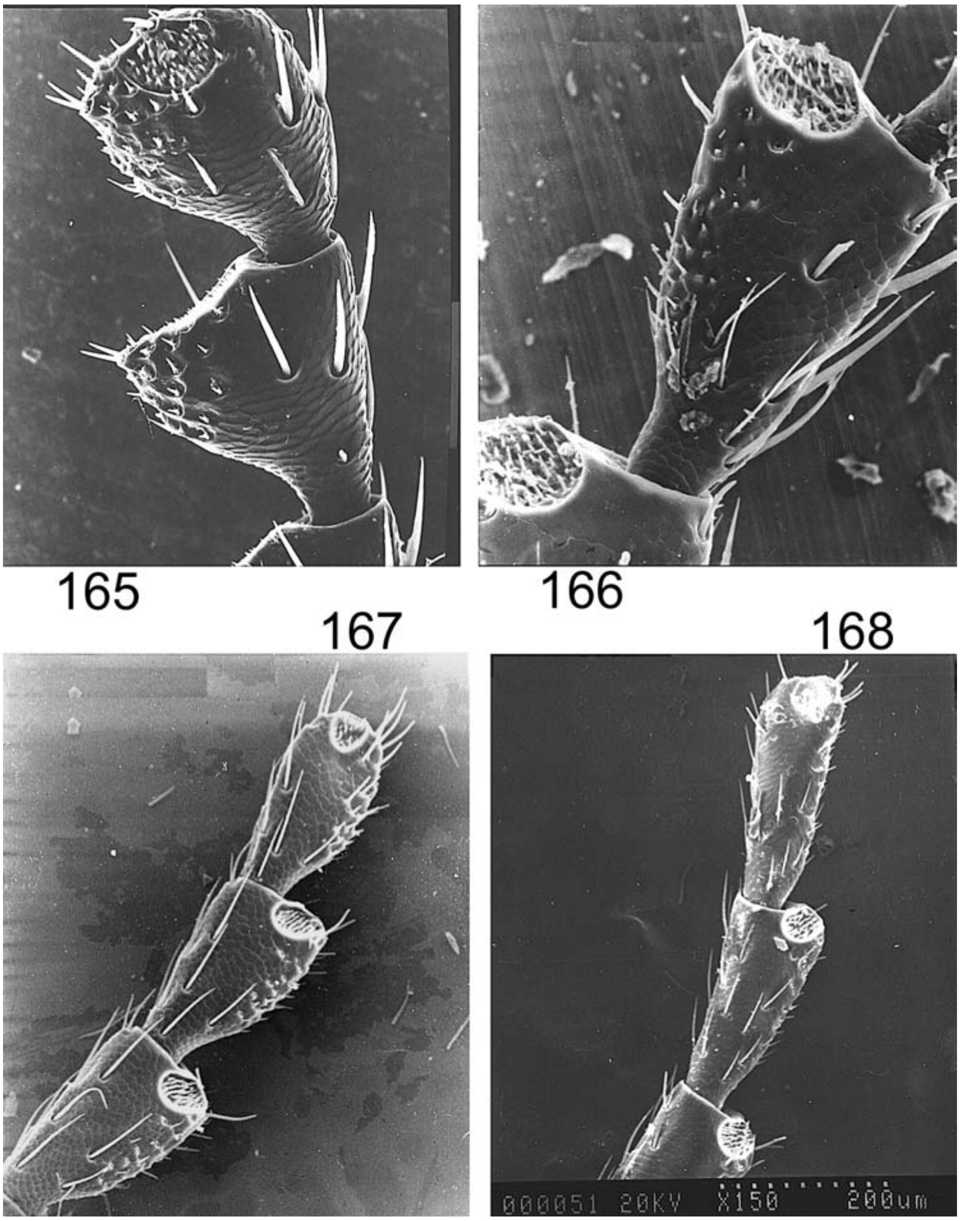
Figs 153–156: *Spectralia*, *Ditriaena*, *Hilarotes*, *Pterobothris*. – 153. *Spectralia purpurascens*: 9–10th, external, ×450; 154. *Ditriaena sexspinosa*: male, 6th, internal, ×350; 155. *Hilarotes nitidicollis*: 9–11th, internal, ×150; 156. *Pterobothris corrosus*: male, 7–11th, internal, ×220.



Figs 157–160: *Pterobothris*, *Nascioides*. – 157–159. *Pterobothris corrosus*: male; 157: 9th, internal, showing apical fossa comprising modified basiconic B4a sensillae, ×3000; 158: 10–11th, external, ×400; 159: 10th, internal, showing the field of shortened uniporous (U1) and modified basiconic (B4c) sensillae, ×2200; 160. *Nascioides enysii*: male, 9–11th, internal, ×200.



Figs 161–164: *Nascio*, *Coomaniella*, *Trigonogenium*. – 161, 162. *Nascio vetusta*, 161: 6–11th, internal, ×80; 162: 10th, internal, showing additional depression ajacent to apical fossa, ×1700; 163. *Coomaniella* (*Coomaniellina*) *taiwanensis*: 6–11th, internal, ×220; 164. *Trigonogenium angulosum*: 9–11th, internal, ×200.



Figs 165–168: *Trigonogenium*, *Neobubastes*, *Neocuropsis*, *Thomassetia*. – 165. *Trigonogenium angulosum*: 10–11th, external, ×300; 166. *Neobubastes australiasiae*: 8–9th, internal, ×300; 167. *Neocuropsis splendens*: 9–11th, internal, ×170; 168. *Thomassetia crassa*: 9–11th, internal, ×300.

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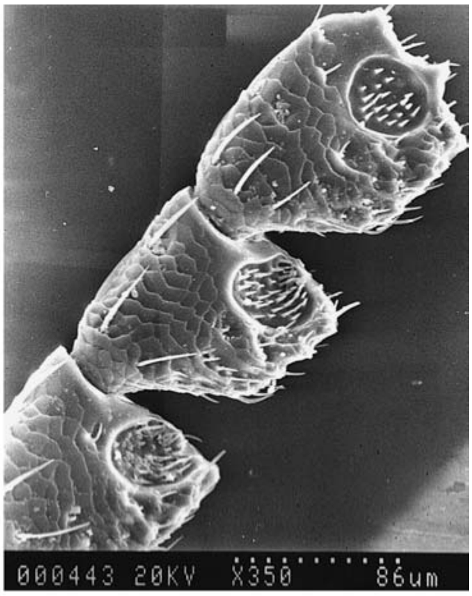

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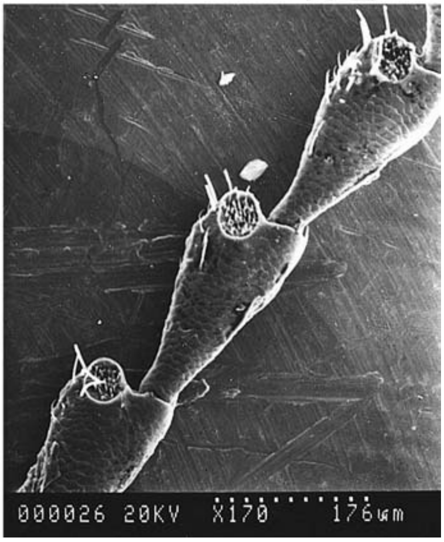
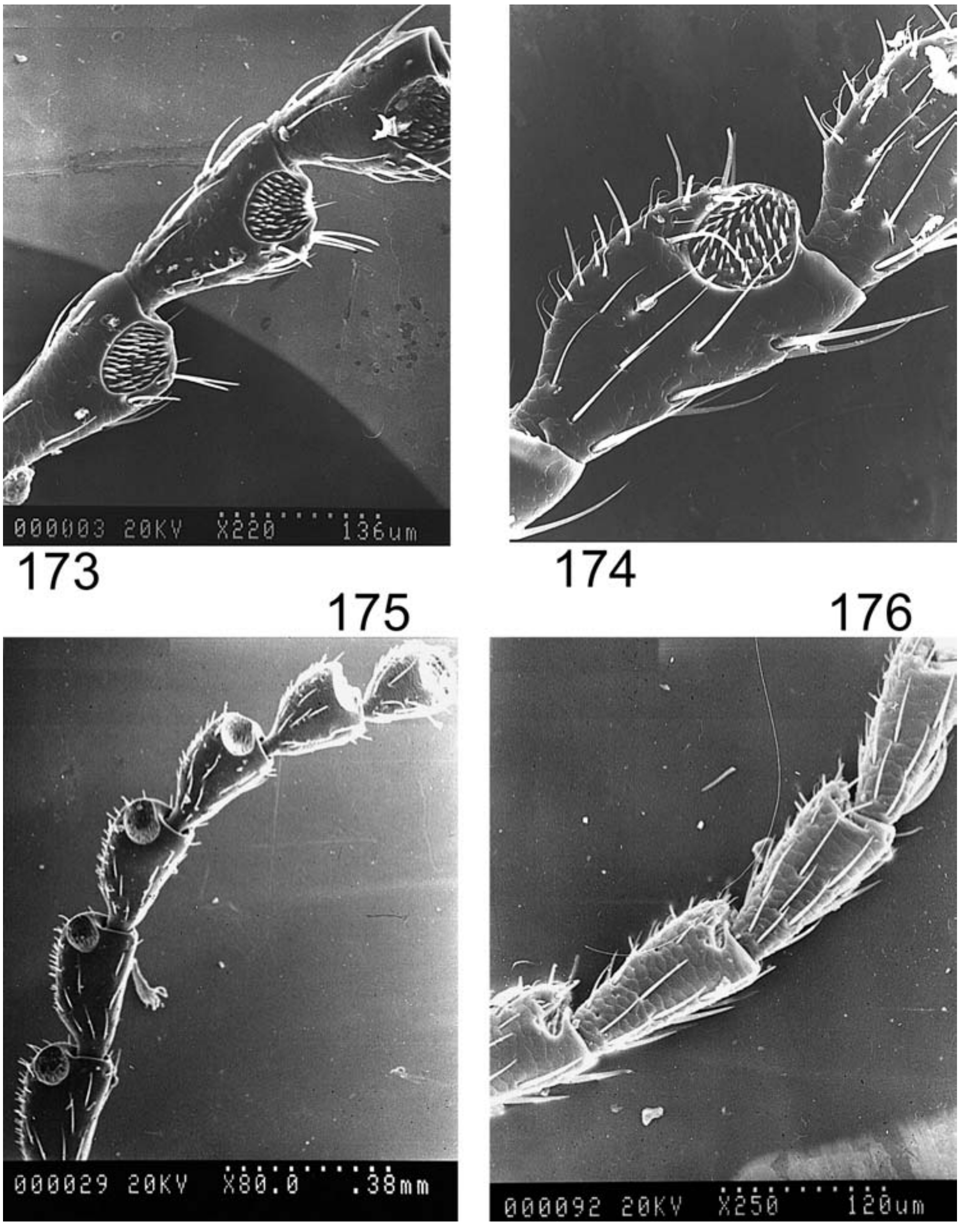
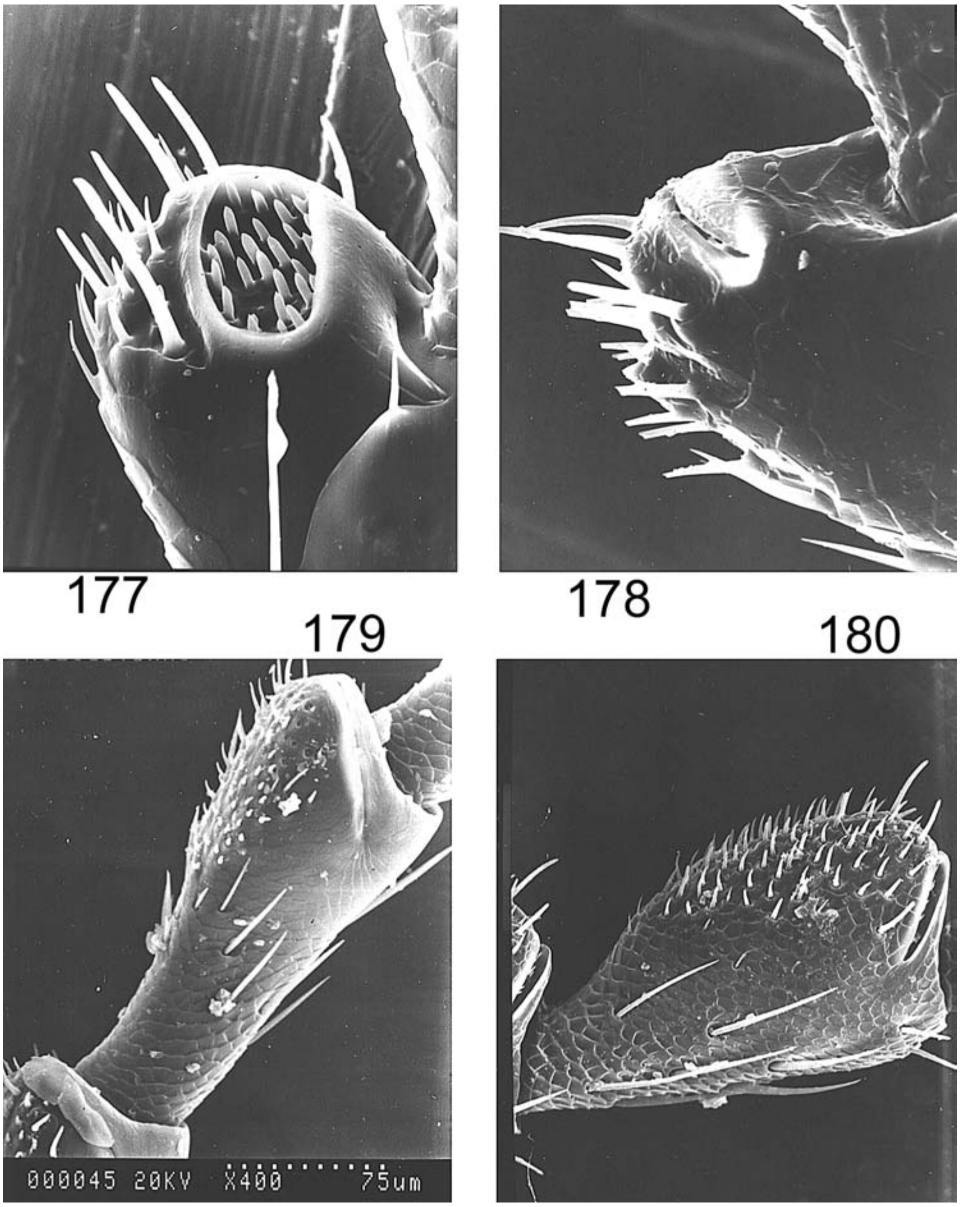


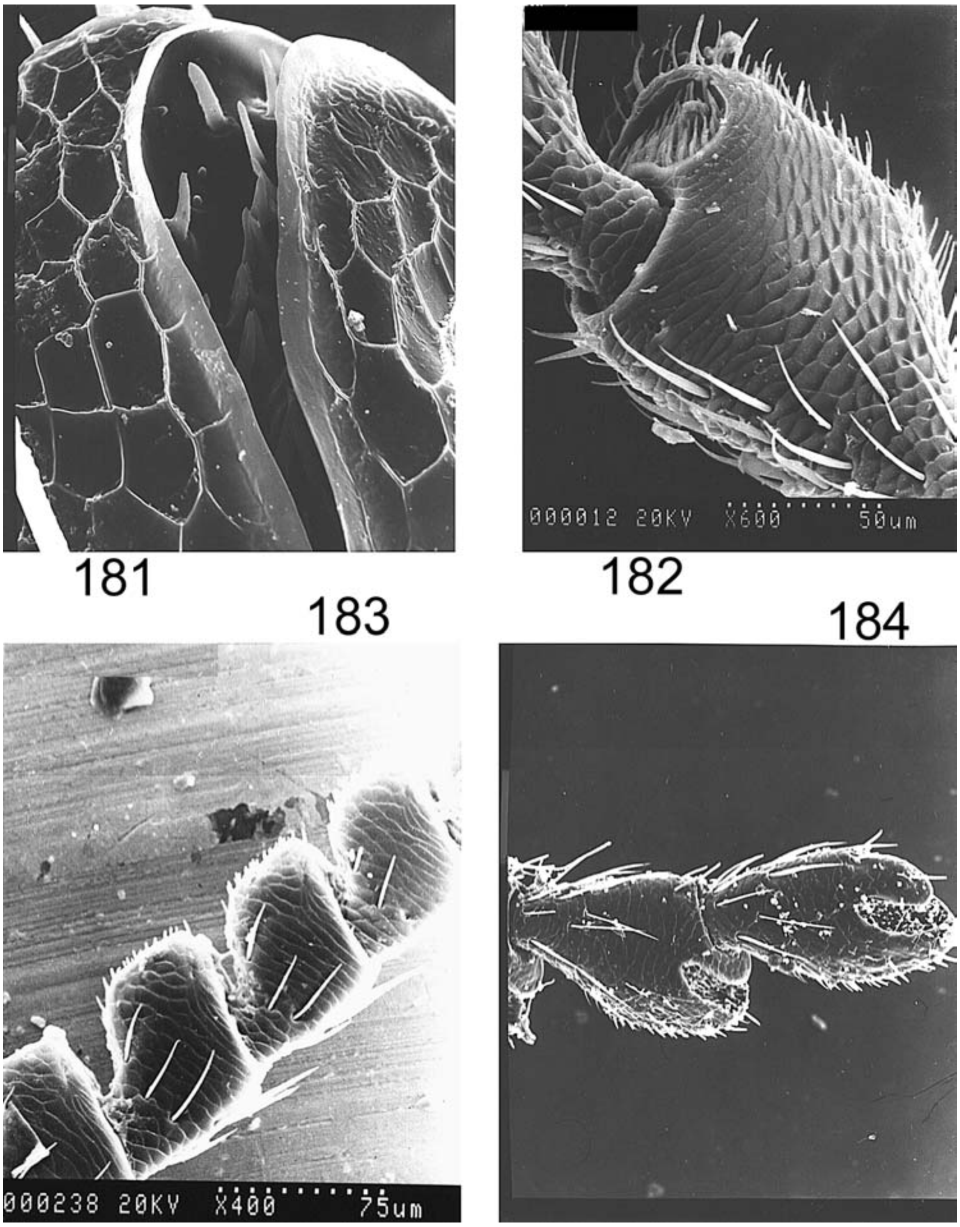
Figs 169–172: *Augrabies*, *Jakovleviola*, *Kisanthobia*, *Aristosoma*. – 169. *Augrabies schotiaphaga*: 6–9th, internal, ×300; 170. *Jakovleviola strandi*: 9–11th, internal, ×350; 171. *Kisanthobia ariasi*: 10th, internal, ×300; 172. *Aristosoma suturale*: 6–8th, internal, ×170.



Figs 173–176: *Philanthaxia*, *Kurosawaia*, *Torresita*, *Theryaxia*. – 173. *Philanthaxia* sp.: 5–7th, internal, ×220; 174. *Kurosawaia* yanoi: 6th, internal, ×300; 175. *Torresita dilatata*: 6–11th, internal, ×80; 176. *Theryaxia suttoni*: 7–10th, internal, ×250.



Figs 177–180: *Neocuris*, *Tetragonoschema*, *Notographus*, *Melobasis*. – 177. *Neocuris* sp.: 6th, internal, ×1000; 178. *Tetragonoschema* sp.: 6th, internal, ×1000; 179. *Notographus sulcipennis*: 6th, internal, ×400; 180. *Melobasis* (s.str.) *gloriosa*: male, 10th, internal, ×300.



Figs 181–184: *Melobasis*, *Anilara*, *Anthaxoschema*, *Cylindrophora*. – 181. *Melobasis*(s.str.) *gloriosa*: male, 10th, internal, showing apical cavity comprising basiconic and campaniform sensillae, ×1500; 182. *Anilara sulcicollis*: 7th, internal, ×600; 183. *Anthaxoschema terraereginae*: 5–9th, internal, ×400; 184. *Cylindrophora maulica*: 10–11th, internal, ×200.

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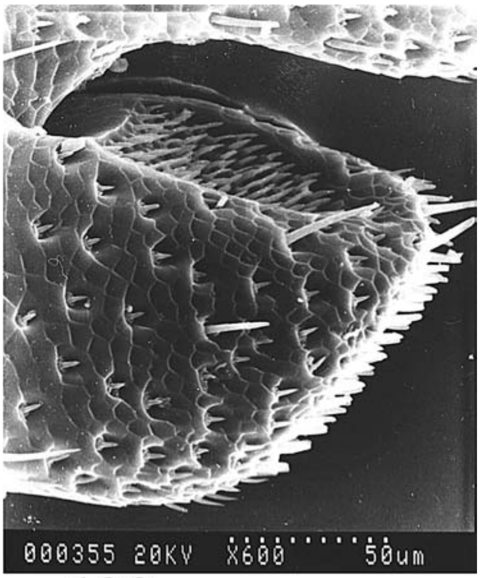

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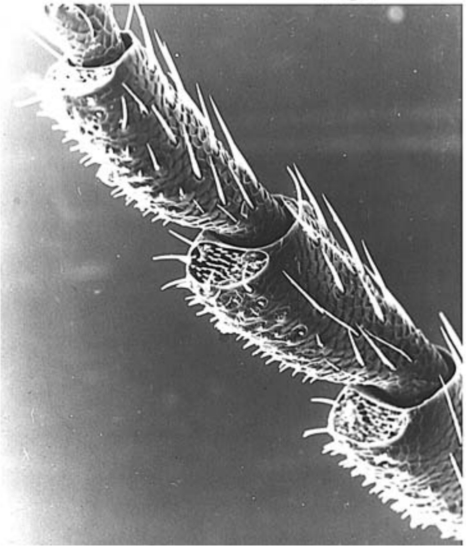

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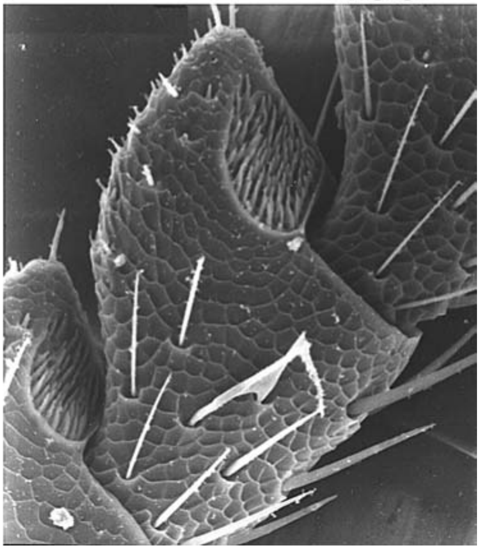

Figs 185–188: *Paracuris*, *Chalcogenia*, *Anthaxia*, *Cratomerus*. – 185. *Paracuris bimaculata*: 7–8th, internal, ×500; 186. *Chalcogenia contempta*: 7th, internal, ×600; 187. *Anthaxia* (s.str) *vittula*: 7–9th, internal, ×300; 188. *Cratomerus* (*Cryptocratomerus*) *iliensis*: male, 5–6th, internal, ×400.

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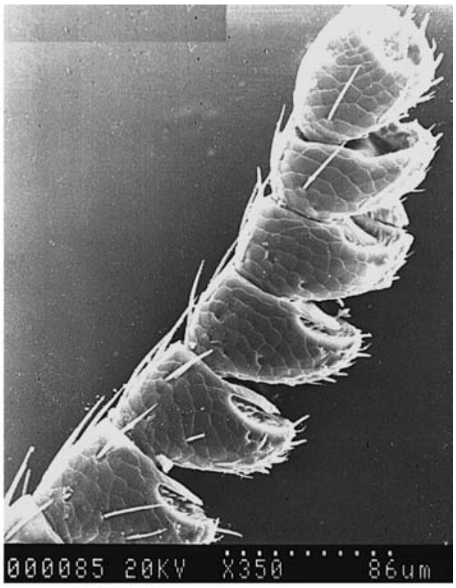

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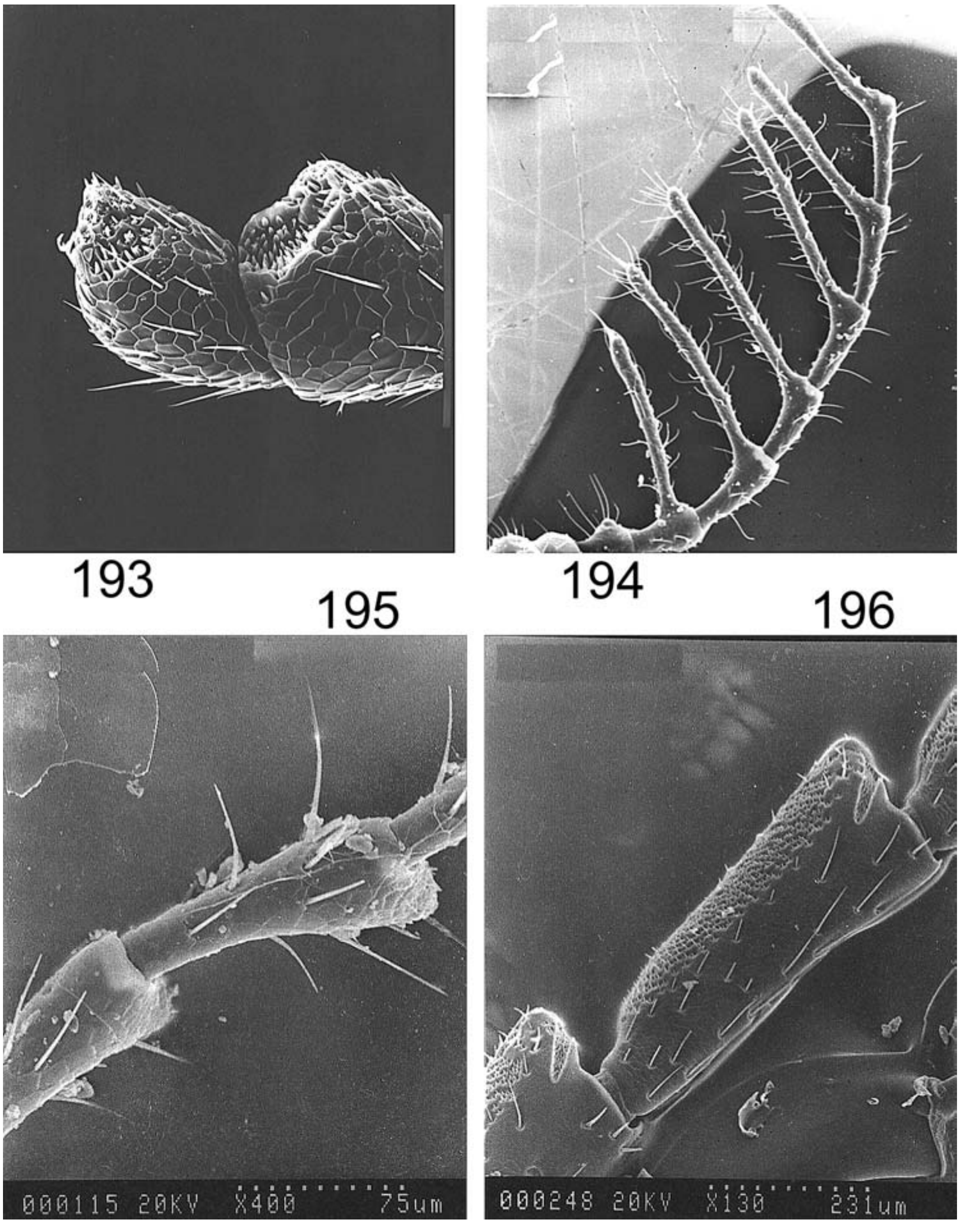
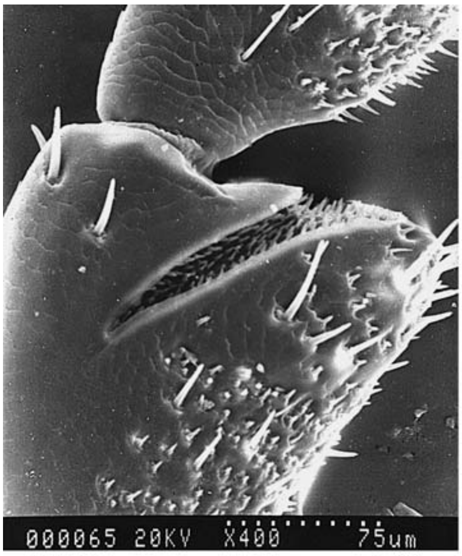
Figs 189–192: *Brachelytrium*, *Brachanthaxia*, *Xenorhipis*. – 189. *Brachelytrium transvaalense*: female, 7–11th, internal, ×250; 190. *Brachanthaxia gemmata*: 6–11th, internal, ×350; 191, 192. *Xenorhipis brendeli*: male, 8th, internal; 191: apical fossa, ×700; 192: biforked male sensillae, ×3000.



Figs 193–196: *Xenorhipis*, *Trichinorhipis*, *Juniperella*. – 193. *Xenorhipis brendeli*: female, 10–11th, internal, ×450; 194, 195. *Trichinorhipis knulli*, 194: male, general view, internal, ×110; 195: female, 5–6th, internal, ×400; 196. *Juniperella mirabilis*: male, 5–6th, internal, ×130.



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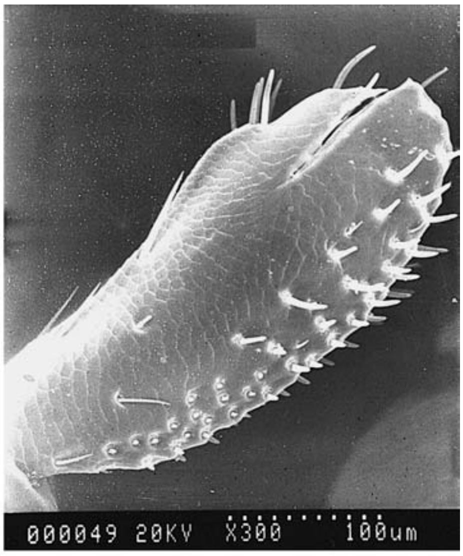

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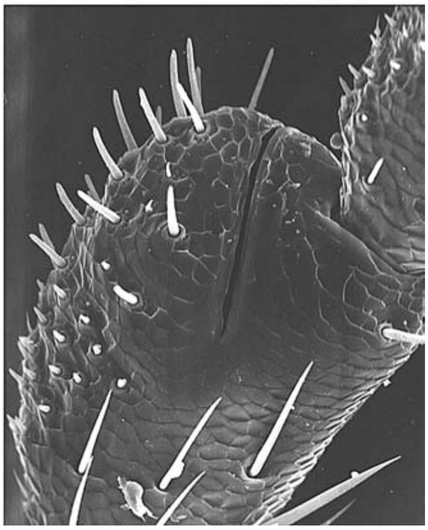
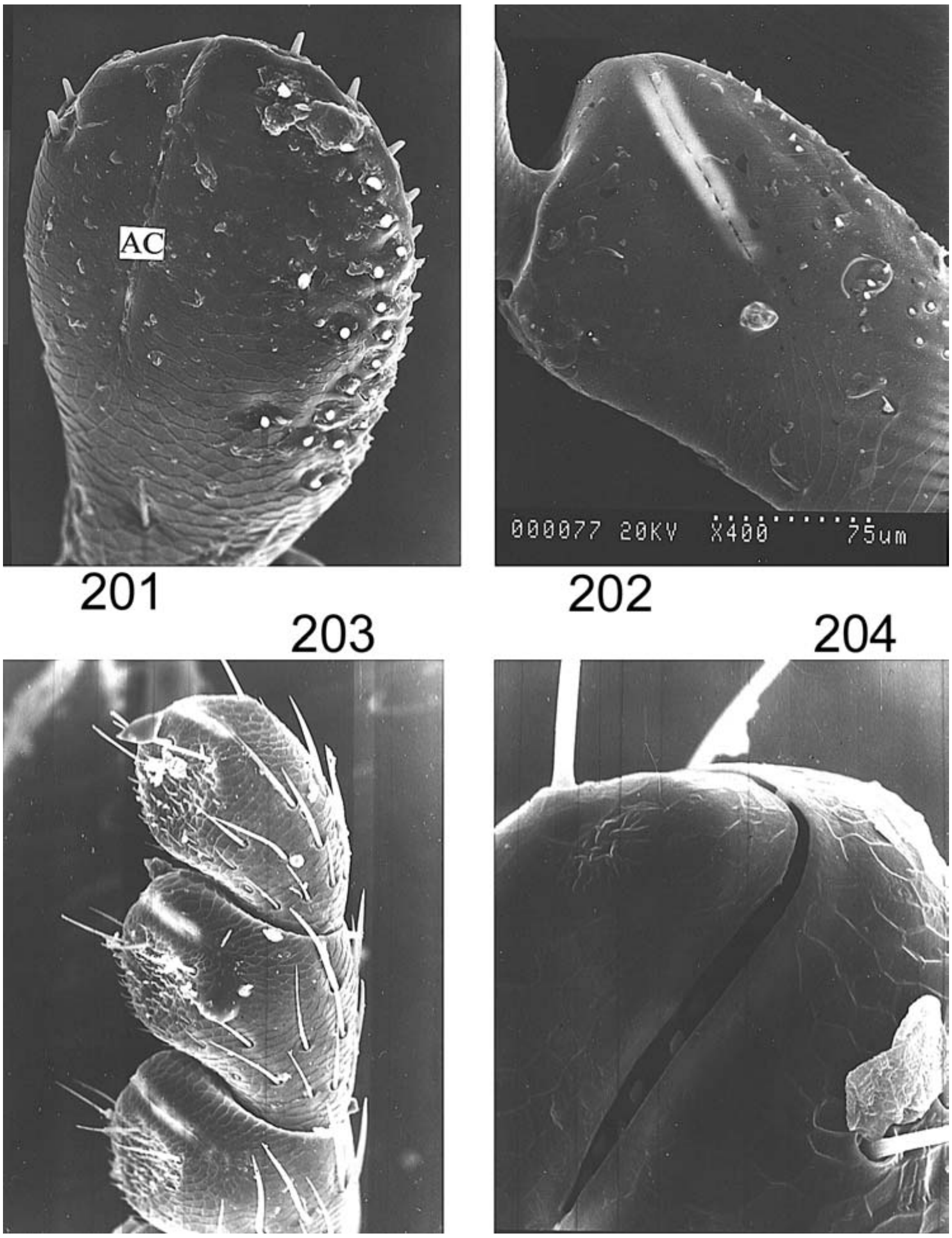
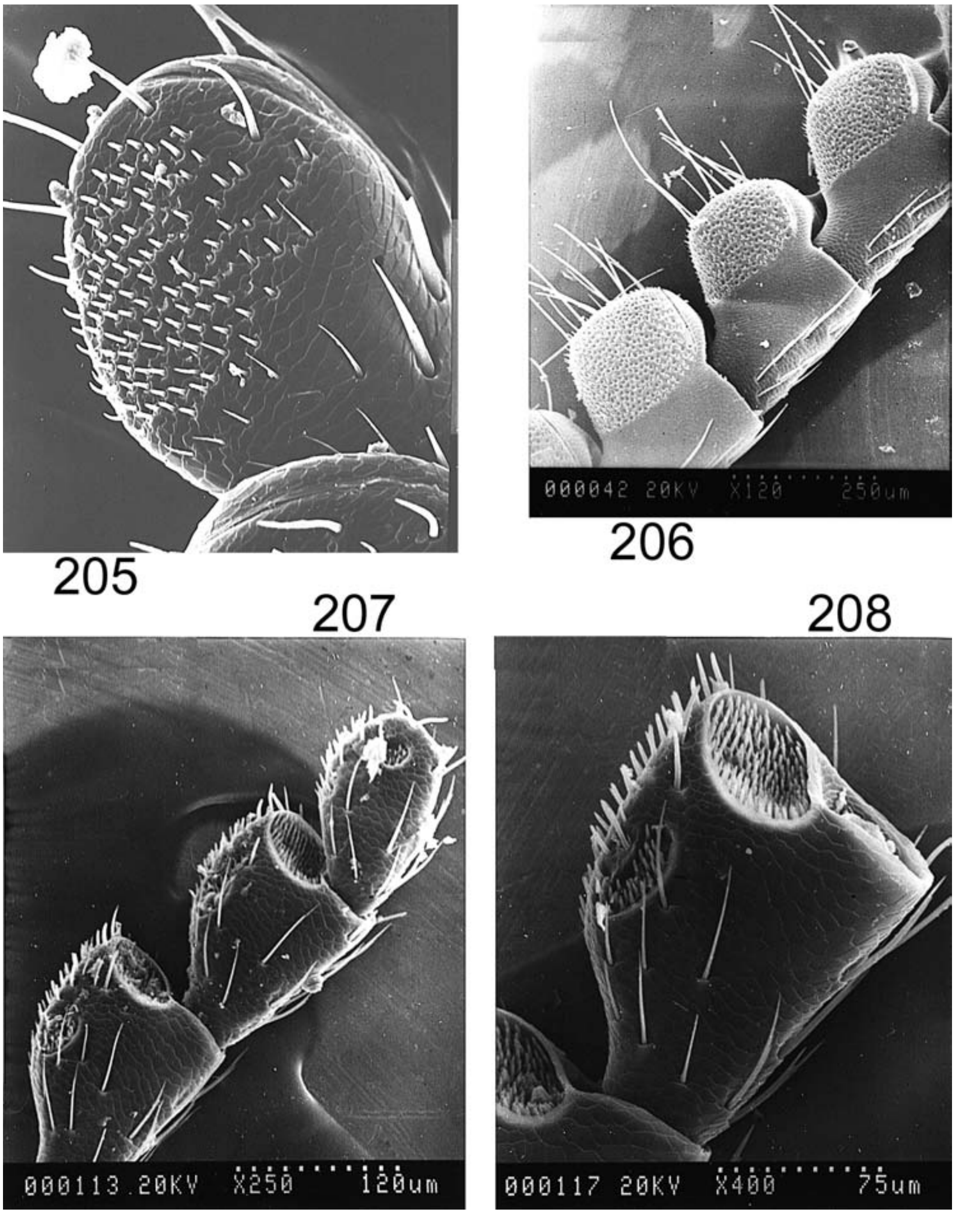


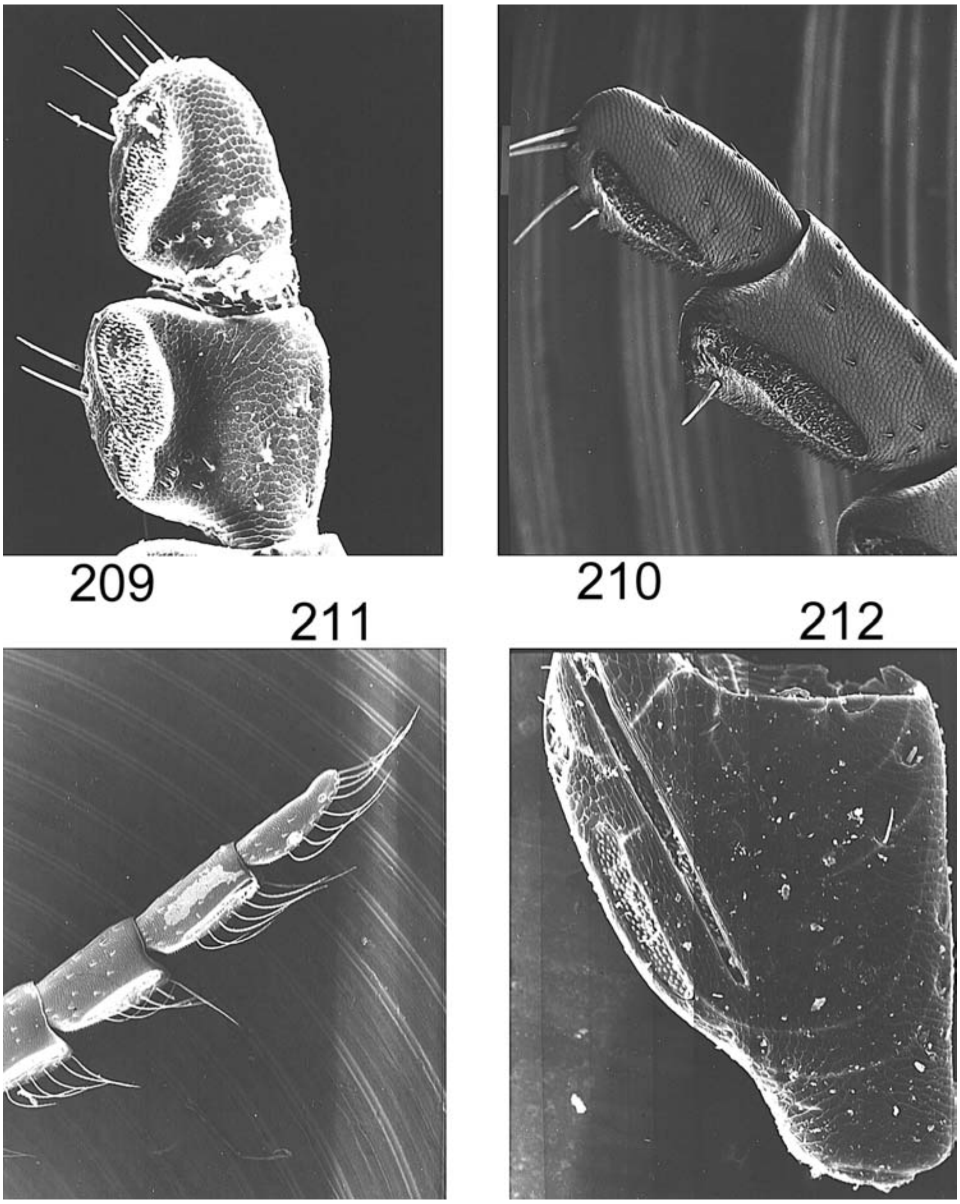
Figs 197–200: *Juniperella*, *Xenomelanophila*, *Phaenops*. – 197. *Juniperella mirabilis*: male, 8th, internal, showing apical cavity containing internal sensillae and sensory field along inner margin, ×400; 198. *Xenomelanophila miranda*: 11th, internal, ×300; 199, 200. *Phaenops guttulata*, 199: 7–11th, internal, ×100; 200: 10th, internal, ×450.



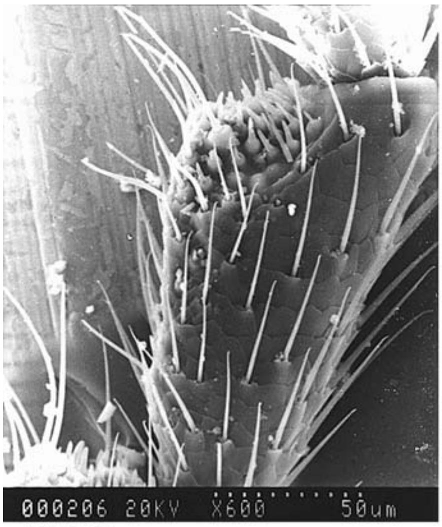
Figs 201–204: *Trachypteris, Merimna*, *Chrysobothris*. – 201. *Trachypteris picta*: 11th, internal, showing apical cavity (AC) completely closed, ×450; 202. *Merimna atrata*: 9th, internal, ×400; 203, 204. *Chrysobothris* (s.str.) *chrysostigma*: female; 203: 9–11th, external, ×200; 204: 11th, internal, showing apical cavity partly open, ×1000.



Figs 205–208: *Chrysobothris*, *Afrobothris*, *Cromophila*. – 205. *Chrysobothris* (s.str.) *affinis*: 10–11th, external, showing apical cavities completely closed and sensory field on inner surface, ×450; 206. *Afrobothris nigrita*: 8–10th, internal, showing apical cavities nearly completely closed, sensory field on internal side, and long ?S8 sensillae along inner margin, ×120; 207, 208. *Cromophila* sp., 207: 9–11th, internal, ×250; 208: 8th, internal, showing apical and lateral fossae, and sensory field along inner margin, ×400.



Figs 209–212: *Actenodes*, *Belionota*, *Pseudactenodes*. – 209. *Actenodes costipennis*: 10–11th, external, ×150; 210. *Belionota* (s.str.) *aenea*: 9–11th, internal, ×150; 211. *Belionota* (s.str.) *sumptuosa*: male, 8–11th, internal, showing apical and lateral fossae comletely confluented into single longitudinal groove, and long serrate male sensillae, ×45; 212. *Pseudactenodes vitticollis*: ?7th, internal, ×200.



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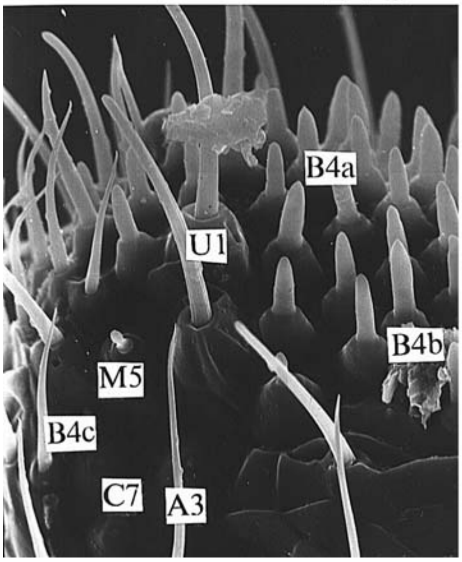

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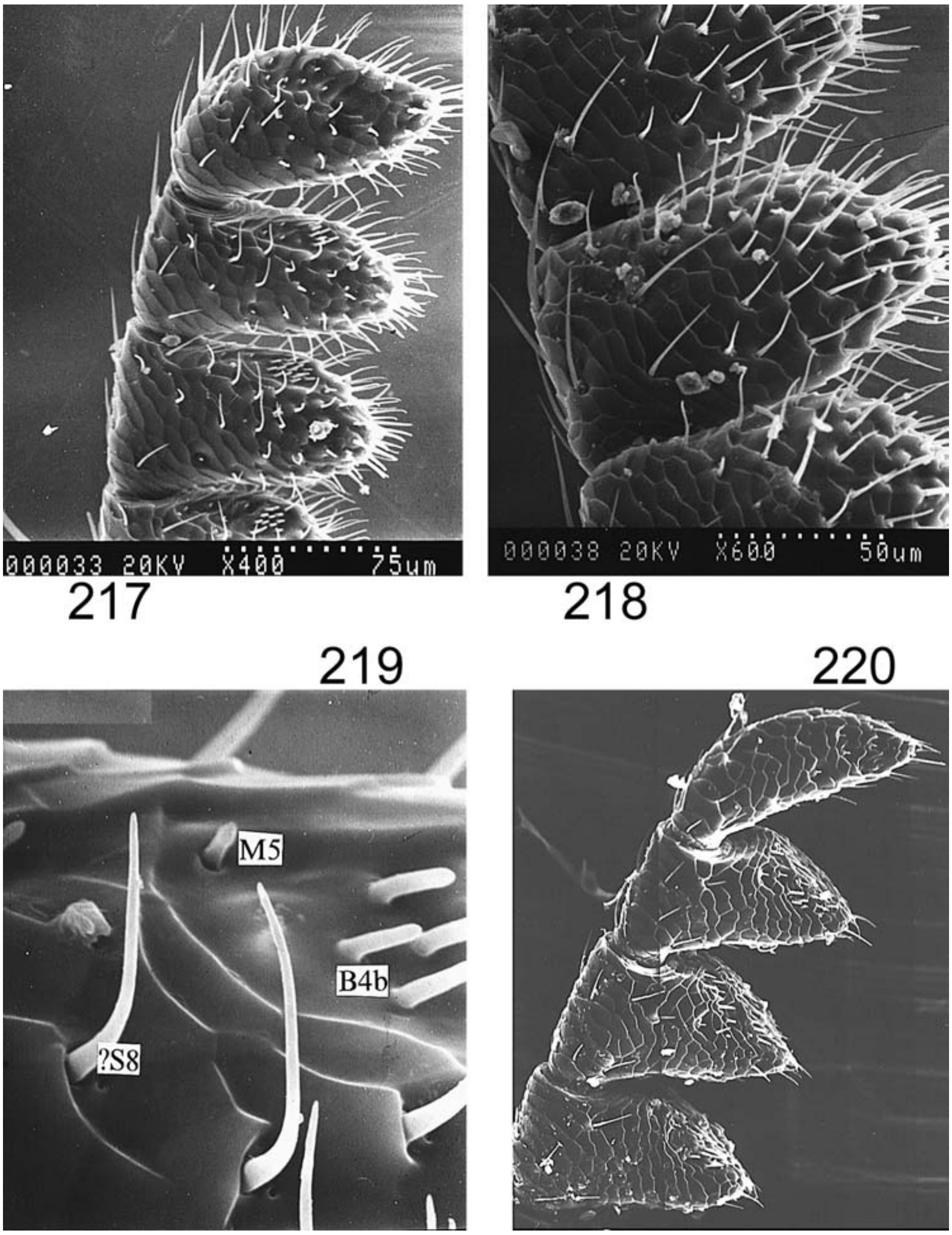
Figs 213–216: *Maoraxia*, *Fahraeusia*. – 213–215. *Maoraxia eremita*, 213: male, 5th, internal, ×600; 214: male, 6th, external, ×500; 215: female, 10th, internal, showing apical sensory zone comprising styloconic uniporous (U1), aporous (A3), basiconic (B4a, B4b, B4c), multiporous (M5), and campaniform (C7) sensillae, ×1500; 216. *Fahraeusia chalcea*: 8–11th, internal, ×80.



Figs 217–220: *Galbella*, 217–219. *Galbella felix*: ?male; 217: 8–11th, internal, ×400; 218: 7–9th, external, × 600; 219: 9th, internal, showing basal part of apical depression comprising basiconic (B4b) and multiporous (M5) sensillae, and external modified (?S8) sensillae, ×3000; 220. *Galbella* sp.: ?female, 8–11th, internal, ×300.