## 1. Introduction

More than 30 excursions to southern Africa have been made since 1992, with the objective of collecting sawflies (Fig.1), a group which has been scientifically neglected in Africa for more than thirty years. Of major importance in this context was the field work of the Biodiversity Monitoring Transect Analysis in Africa (BIOTA) from 2000 to 2010, especially phase III from 2007 to 2010. The BIOTA-Africa Project comprised over 30 subprojects in a number of countries in eastern, western and southern Africa. The research sites of the BIOTA-Southern Africa Project follow rainfall gradients from dry forest habitats through arid savanna to the winter rainfall zones in Namibia and South Africa (**Chapter 2: The study area**). The central theme of this project was to document biodiversity in space and time.


**Fig. 1.** *Xenapates gaullei* (Konow, 1896). Its larvae feed on different species of Poaceae in western Africa. (Photo by G. Goergen)

Parallel to this, numerous unidentified specimens in South African and Namibian collections were also examined. It became clear that satisfactory identification could not be achieved through use of the existing literature and that these problems could only be solved by revision of the affected genera. Furthermore, during initial attempts at field work, experience gained from the collection of these genera in central Europe failed to lead to comparably good results in southern Africa. Even for practised entomologists, sawflies are difficult to find, especially in the arid and semi-arid parts of southern Namibia and the Northern Cape and Western Cape provinces of South Africa. To optimise collecting results, it was judged necessary to obtain more precise data on the habitats and climatic conditions that best suit Symphyta in these regions (**Chapter 5: Material and methods**).

Comprehensive taxonomic results and ecological information acquired over the last 20 plus years on the Symphyta fauna of southern Africa, which now comprises 14 genera and 55 species for the study area, constitute the main component of this monograph. This work provides a baseline for further research on Symphyta of the Afrotropical Region.

This publication was first envisaged as a “Field guide of the Symphyta of Namibia and the western parts of South Africa”, comparable with numerous available books on the butterflies, dragonflies and conspicuous groups of beetles of this region.

However, during development of the manuscript doubts arose as to whether the Symphyta could ever attract the interest of such a wide audience.

The sawfly fauna of this region seems to be very poor in number of species. Especially the dry biomes, Succulent Karoo and Nama Karoo, appear to offer rather unsuitable conditions for the successful completion of the life cycle of these phytophagous insects, whose larvae feed externally on plant foliage. Among other factors, the poor knowledge of sawflies from this region can be explained by the fact that thorough field collections specifically targeting this group have not been undertaken in these arid and semiarid habitats.

By comparison, sawflies are far more abundant in the humid areas of northern Namibia as well as the montane areas (Afromontane Region) of the South African provinces of KwaZulu-Natal, Mpumalanga, Limpopo and North-West (Koch 2005a, b). Specimens are mainly represented by the black/yellow coloured species that are more visible in this habitat and hence easier to collect because they stand out with their relatively quiet flight against the green or dark background of lusher vegetation.

The question was raised as to whether a careful observer of nature would be interested in these relatively inconspicuous fly-like insects. And if so, the feasibility of achieving an identification in the field without capturing and examining the specimen under a microscope.

Therefore it was decided to create a stable taxonomic base to facilitate further research on Symphyta in southern Africa. This resource will make it possible to accurately determine the species present in the study region (**Chapter 2: The study area**) enabling the discovery of new species.

It is clear from the relevant literature (Prinsloo 1985, Scholtz & Holm 1985, Picker *et al*. 2004) that sawflies have been largely neglected from the perspective of their diversity, as well as ecology and biology in southern Africa. Only the genera *Arge* and *Athalia* are somewhat well represented in the region. The indigenous fauna is supplemented by four introduced species from Europe: *Caliroa cerasi* (Linnaeus, 1758) (Pear-slug), is a pest on fruit crops, the other, *Fenusa dohrnii* (Tischbein, 1846), lives on alder (*Alnus*), *Nematus oligospilus* Förster, 1854, has been reported on cultivated willows (*Salix*) and finally *Sirex noctilio* Fabricius, 1793 (European woodwasp, Sirex woodwasp), a pest in pine plantations. This deficit in the knowledge of the region's sawfly richness is even more remarkable when one considers that in southern Africa an estimated excess of 100 valid species is expected. The existing fragmented literature only enables inaccurate and partial determination of the species

This book fills this gap in our knowledge by providing a resource to initially identify all genera of southern Africa and with the use of additional current literature to

identify the species. The treatment also highlights the genera in the Afrotropical Region requiring taxonomic revision.

A synopsis of sawfly diversity in each of the region's biomes is presented including data on the species' habitat preferences. In addition, information on their flight seasonality and distribution throughout the Afrotropical Region as well as a compilation of the available information on host plants and other biological data are provided.

The remaining chapters deal with the ecological function of Symphyta in ecosystems and their importance as plant pests. Based on examination of historical material preserved in museum collections and experience gained during field research over the last 20 years, an assessment of potential extinction threats to species and the requirements for their future conservation are discussed.

## 1.1 Sawflies as part of the ecosystems

Sawflies are very rarely observed in the biomes of the study area, especially in the arid regions of the winter rainfall zone. Even in the other more mesic biomes (Thornbush Savanna and Woodland Savanna) the population density of Symphyta appears to be low and therefore we can only speculate about their function in these ecosystems.

Adult sawflies are occasionally found in abundance on flowers, with a preference for Asteraceae, where they feed on pollen and possibly also nectar (**Chapters 6: Life cycle; 7: Host plants**). We know very little about relationships of adults to certain plant species based on limited observations in the field. Very rarely larvae have been observed, however, no females have been observed laying eggs. In **Chapter 7: Host plants** as well as in the species treatments, recorded observations are presented for the various species providing a base for further ecological studies.

The relatively low abundance of Symphyta in southern Africa in combination with high predation pressure from ants, spiders, birds, small mammals and parasitoids on larvae and adults means that they may not play a significant role in food webs. Further ecological studies are still required to determine the exact role that they play in ecosystem functioning.

## 1.2 Importance of sawflies as pests of agricultural crops

Little is known about endemic sawfly species as pests of crop plants in southern Africa (Visser 2009). A few larvae of *Athalia* species have been recorded to feed on cabbage and turnips (Brassicaceae) and some introduced pests of decidious fruit are known such as the Pear-slug (*Caliroa cerasi*) (Visser 2009). Since even the *Athalia* species imaged by Visser (2009) were unidentified, this once again demonstrates the importance of this book for the identification of species

allowing for implementation of effective measures to protect crops. *Arge taeniata* (Klug, 1834), known as the Pelargonium sawfly, occurs as a pest in *Pelargonium* plantations in the winter rainfall area of Western Cape Province (Prinsloo 1985).

*Sirex noctilio*, an introduction from Europe, is an increasingly important pest in the timber industry (Fig. 183) (van Noort & Picker 2011). *Nematus oligospilus* Förster, 1854, has been recorded as feeding on willows (Urban & Eardley 1995, 1997).

In equatorial Africa, the importance of endemic Symphyta as pests on crop plants is more significant. This applies particularly to several *Athalia* species on Brassicaceae (Benson 1962) and *Xenapates* species (Fig. 1) on Poaceae and Commelinaceae (**Chapter 7: Host plants**). The identification keys presented here, especially to the genera, are very useful for the rest of Africa. Most African genera are identifiable using these keys, and recommendations to additional specific literature are provided.

## 1.3 Destruction and conservation of natural habitats

The spectrum of species, covered in this treatment includes numerous recently described species, especially from the winter rainfall area. Some of these species were discovered in small numbers in very old collections in different South African

**Fig. 2.** Destruction of the coastal vegetation by urban land use expansion across the Cape flats near Muizenberg adjacent to False Bay south of Cape Town (Western Cape Province). (Photo by S.van Noort)

museums and were described as part of the preparation of this monograph (Koch 2006a, Koch & Goergen 2010, Koch & Eardley 2011).

Despite intensive field work over several years, very few new records of these species were assimilated. The same is true for species described last century, many of which have never been recollected since their original description. The documentation of all species, however, is indispensable for assessment of species richness and diversity prior to habitat transformation and climate change.

The disappearance of sawfly species from the study area and other areas of South Africa is probably a result of changes in space and time relating to shifts due to changes in ecological conditions, including the destruction of the original habitats over the past 100 years. This is especially true for the metropolitan area of Cape Town that has expanded northwards and eastwards towards Paarl, Stellenbosch and Somerset West (Fig. 2).

Destruction of natural habitats has also occurred as a result of extensive crop farming and stock farming (Figs 3, 4), as well as wine and citrus cultivation (Fig. 5), particularly in the Fynbos Biome. Agricultural land use has strongly reduced and split up the original primary vegetation, especially in the Coastal Fynbos (Fig. 6). The same phenomenon is caused by the increasing expansion of the timber industry in

**Fig. 3.** Destruction of natural habitats by extensive agricultural land use. View from the Gydo Pass near Ceres towards the Grootwinterhoek Nature Reserve (Western Cape Province). (Photo by F. Koch)

**Fig. 4.** Extensive crop farming south of Swellendam (Western Cape Province). (Photo by F. Koch)

**Fig. 5.** Extensive cultivation of citrus and wine near Citrusdal (Western Cape Province) (Photo by F. Koch)

**Fig. 6.** View from the border of De Hoop Nature Reserve (Western Cape Province). Agricultural land use dominates the landscape. The primary vegetation of the Coastal Fynbos vegetation type only persists in island patches (these archipelago-like remnants are visible in the background). (Photo by F. Koch)

areas of Mountain Fynbos (**Chapter 2: The study area**). In many areas, extensive afforestation has destroyed most of the natural vegetation, and large tracts of land have been severely invaded by alien plants (*Pinus* and *Eucalyptus*). The primary vegetation remaining is in only poorly accessible habitats or in protected areas.

With this unbridled growth of urban and rural land use, the natural habitat of many sawfly species is being destroyed, and they can only survive in the protected areas. This is especially true for species with small distribution areas, and it seems probable that some of these species are now extinct (**Chapter 9: Systematic presentation**). From a conservation point of view, it is critical to document the sawfly fauna in the remaining original habitats.

In this respect, the data presented here has value in supporting the creation of large conservation areas in the unique Fynbos Biome.

## 2. The study area

The study area (Fig. 7) includes the entire territory of Namibia including the Caprivi Strip. In the South it extends over Northern Cape Province and Western Cape Province to the Indian Ocean. The South African part of the study area is delimited in the east by the 21st longitude.

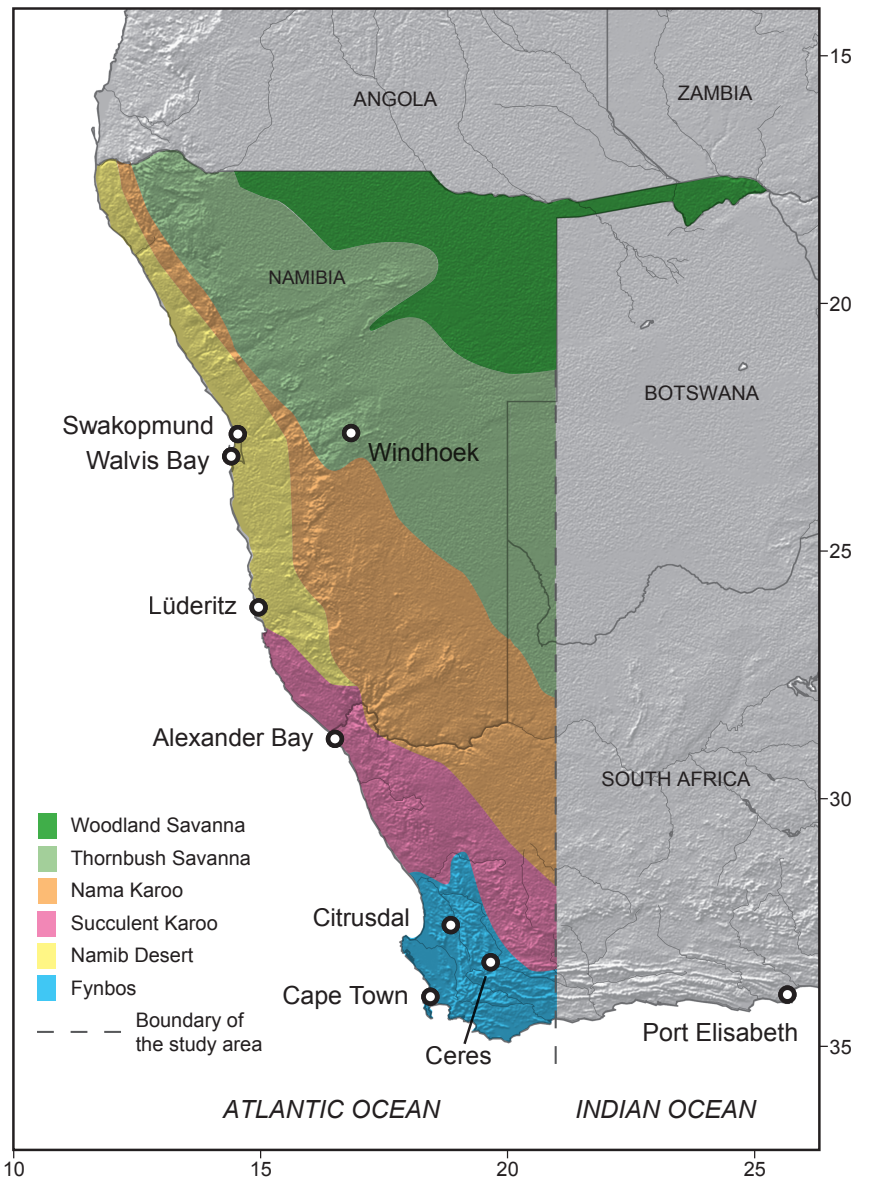


**Fig. 7.** The study area with its biomes, modified after Jäschke & Langner (2010).

This area includes nearly the whole winter rainfall area of southern Africa (Fig. 8). The region of the winter rainfall encompasses an area approximately positioned west of a line extending from Swakopmund on the Atlantic coast of Namibia to Mosselbaai on the Western Cape coast of South Africa. The northern part of this line is characterized by the Succulent Karoo Biome and the southern part is dominated by the Fynbos Biome.

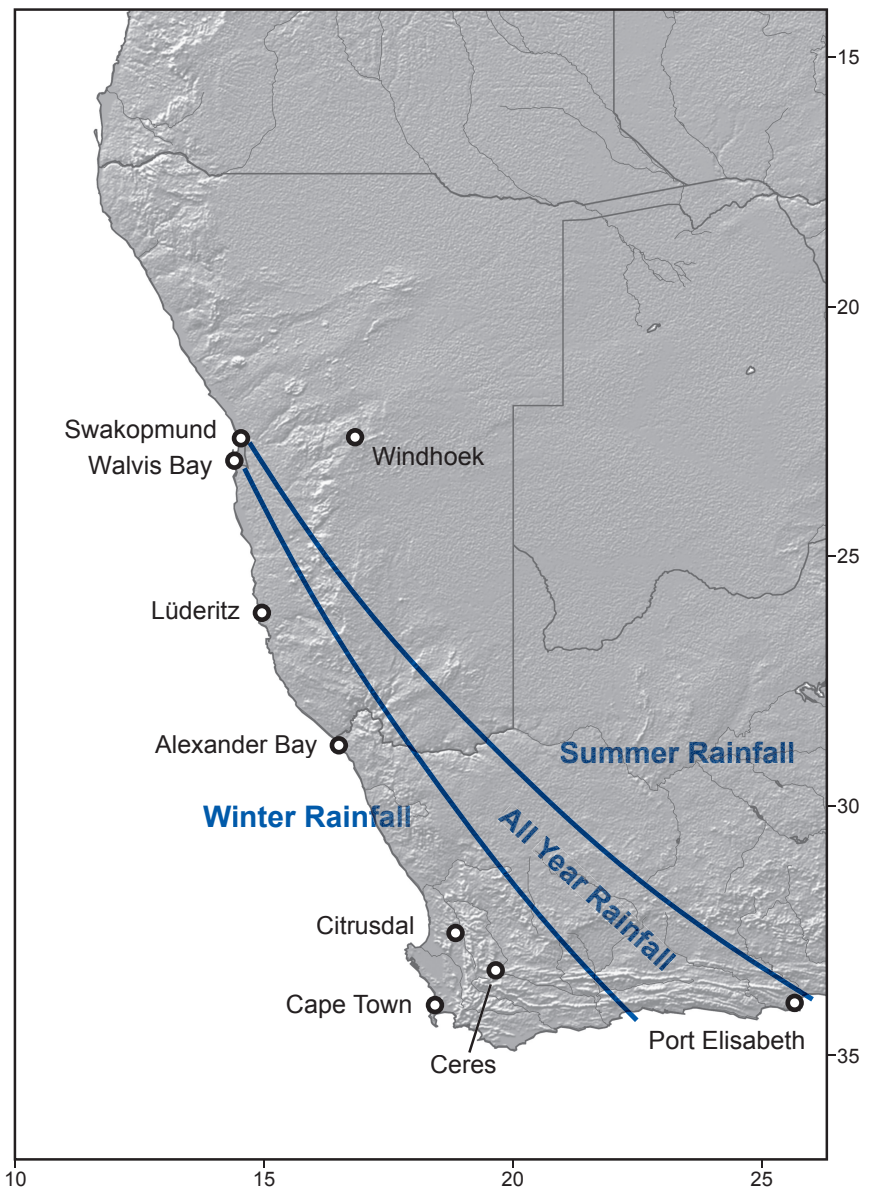


**Fig. 8.** The rainfall zones of the study area.

## 2.1 Biomes of the study region

### Woodland Savanna Biome

The Woodland Savanna Biome (Fig. 7) is characterised by dry woodlands dominated by tall trees and relatively sparse understory vegetation. This biome covers large parts of the African continent. In Namibia it is situated in the north-east area bordering Angola and Botswana and receives much higher annual rainfall than the rest of the country. There is a typical summer rainfall climate, with an average annual rainfall of about 500 mm. This higher rainfall allows for the typical, mainly non-commercial land use structure in the area, which consists of rain-driven subsistence crop activities during high rainfall years (Jürgens *et al*. 2010).

The centre of the Woodland Savanna Biome is characterised by the extensive sand plateau of the Northern Kalahari, and possesses linear east-west orientated structures which are remnants of a prehistoric dune field. These dunes have since been eroded, sometimes to a level lower than the more loamy remnants in the adjacent inter-dune valleys. The deep sands of the Kalahari Plateau are stabilised by an open broad-leaved woodland (Figs 9, 10).

The so called Caprivi Strip in the Northeast of Namibia in the Woodland Savanna Biome is characterised by the large streams Okavango River in West Caprivi and

**Fig. 9.** The Woodland Savanna Biome in northern Namibia (Mahango Game Reserve). (Photo by F. Koch)

**Fig. 10.** An impression of the Woodland Savanna Biome of the Northern Kalahari (Kalahari Basin, north-eastern Namibia). (Photo by J. Deckert)

**Fig. 11.** The wetlands of the Woodland Savanna Biome near Schuckmannsburg on the Zambezi River in the Eastern Caprivi (north-eastern Namibia). (Photo by F. Koch)

the Zambezi River and Kwando River in the East Caprivi with their riverine forest. Additionally, the Kwando River, the Linyanti River and the Chobe River in the East Caprivi area are adjoined by extensive wetlands (Fig. 11).

Furthermore, numerous ephemeral waters (dry rivers and pans), which are dependent on unpredictable rainfall are located in this biome. Pans are natural depressions that fill with water after periods of heavy rain.

**Thornbush Savanna Biome**

These savannas, which are characterised by an extensive cover of grasses, with scattered trees (Fig. 12), cover almost one-third of the land surface of the earth and 40% of Africa. In southern Africa, savannas are home to growing human populations and most people depend on the ecological resources these systems offer. The main landuse practices in the Thornbush Savanna Biome (Fig. 13) are farming with cattle and – increasingly so – game for trophy hunting (Jürgens *et al*. 2010).

The Thornbush Savanna Biome (Fig. 7) belongs to the summer rainfall area. It involves numerous ephemeral waters, which are filled with water after periods of heavy rain. This applies for example to the famous Etosha Pan in the North of Namibia and large rivers that rise here and flow into the Atlantic Ocean.

**Fig. 12.** A game hunting farm north of Windhoek, near Okahandja in the Thornbush Savanna Biome (Namibia). (Photo by J. Deckert)

**Fig. 13.** Cattle farm “Erichsfelde” in the Thornbush Savanna Biome near Okahandja (Namibia). (Photo by J. Deckert)

**Fig. 14.** The Kaokoveld in the Thornbush Savanna Biome in north-western Namibia. (Photo by J. Deckert)

The average altitude is about 1200 m a.s.l. (Fig. 14), and ranges from 1800 m to 2479 m in the mountainous highlands west of Windhoek commonly known as “Khomas Hochland”.

Biogeographically, the Waterberg area in Namibia is a mix of both Woodland Savanna Biome and Thornbush Savanna Biome. In the map of biomes (Fig. 7), the Woodland Savanna Biome overlaps with a very narrow tip the Waterberg Plateau Park. The geobotanical elements of the Woodland Savanna Biome are particularly located on the plateau, and the area around the mountain is characterized by typical plants of the Thornbush Savanna Biome. The material mentioned from the Waterberg in this book was collected somewhat above or directly at the foot of the mountain and therefore most of these habitats belong to the Thornbush Savanna Biome.

**Nama Karoo Biome**

The Nama Karoo Biome (Fig. 7) is characterised by sparse vegetation composed of shrubs and grasses, which is comparatively species-poor and does not comprise many local endemics. This semi-desert shrubland with extensive areas of rocky outcrops and limited grass cover belongs to the summer rainfall area, however the annual rainfall is less than 400 millimetres (Fig. 15).

**Fig. 15.** The Gondwana Canyon Park (southern Namibia) in the Nama Karoo Biome with endemic quiver tree (*Aloe dichotoma* Masson). (Photo by J. Deckert)

**Fig. 16.** The Brandberg Massif in the northern Nama Karoo Biome (western Namibia). (Photo by W. Mey)

Due to much lower annual rainfall than the adjacent Thornbush Savanna Biome, taller trees are scarce and largely restricted to mostly dry riverbeds. There is little natural permanent surface water to be found in the Nama Karoo.

The only permanent water-bearing stream is the Gariep [Oranje] River on the border to South Africa. The Brandberg Massif, the highest mountain range in Namibia, is located in the North of the biome (Fig. 16). Farming with sheep and goats is the typical landuse in this biome.

**Succulent Karoo Biome**

The Succulent Karoo Biome (Fig. 7) is located near the west coast in the winter rainfall area. This biome is renowned as a world centre of endemism and biodiversity. More than 600 plant species are endemic to the Succulent Karoo Biome (Figs 17, 18), but local plant endemism is far less. For example a total of 184 plant species was recorded in the area south-east of Lüderitz (Burke 2004).

The biodiversity of the Succulent Karoo Biome is primarily driven by mild climatic conditions. These conditions are characterized by winter rainfall with mild temperatures during the growing season, and additional water supply from fog

**Fig. 17.** The Koeroegapvlakte of the Richtersveld National Park in the Succulent Karoo Biome, Northern Cape Province. (Photo by J. Deckert)

**Fig. 18.** The Succulent Karoo Biome west of Kamieskroon. (Photo by J. Deckert)

and dew. Further factors such as soil type, and bioturbation by termites and small mammals, results in typically patchy habitat conditions and related small-scale patterns of vegetation and phytodiversity. Due to the low annual rainfall, the carrying capacity of the rangelands is very low and the rich biodiversity is threatened by habitat transformation and subsequent species loss due to unsustainable landuse (overgrazing, ploughing, and mining) as well as projected climate change (Jürgens *et al*. 2010).

**Fynbos Biome**

The Fynbos Biome (Fig. 7) belongs also to the winter rainfall area with a comparatively high rainfall season extending from end of May to end of August. This biome is floristically characterized by an extremely dense vegetation dominated by hard-leaved shrubs, small trees, and grass-like growth forms. The biome is part of the Cape Floristic Region, a global biodiversity hotspot confined to the south-western part of South Africa. A combination of topographic, edaphic, and climatic gradients and the frequent occurrence of fire have been suggest as the main driving forces of speciation in the Fynbos Biome, resulting in a great variety of vegetation types (Jürgens *et al*. 2010).

**Fig. 19.** The Mountain Fynbos in the Bokkeveld Mountains (Western Cape Province). (Photo by M. Uhlig)

**Fig. 20.** The Dune and Coastal Fynbos vegetation type in De Hoop Nature Reserve (Western Cape Province). (Photo by F. Koch)

The low lying areas in particular have been extensively transformed due to agriculture, urbanisation, and invasion, and once continuous habitats have been reduced to small, fragmented islands surrounded by transformed land (Fig. 6) (**Chapter 1.3: Destruction and conservation of natural habitats**).

The Fynbos Biome is subdivided into the Mountain Fynbos (Fig. 19) extending north and south of Citrusdal and in the area of Ceres, and the Coastal Fynbos (Fig. 20) situated parallel to the southern and western coastlines of the Cape of Good Hope.

Additionally, different major vegetation types are defined, for example West Coast Strandveld vegetation type “Cape Flats Dune Strandveld” (Mucina & Rutherford 2006) (Figs 49, 56), Sandplains Fynbos, Sandstone Fynbos (Fig. 120) and Renosterveld. All vegetation types are highly endangered by fragmentation, transformation, and invasive alien plant species (Veste & Jürgens 2004, Jürgens *et al*. 2010) (**Chapter 1.3: Destruction and conservation of natural habitats)**.

**Namib Desert Biome**

The Namib Desert Biome (Fig. 7) covers a large area of Namibia and stretches along the western seaboard into South Africa. It is characterized by extremely low rainfall (less than 50 millimetres per annum) and an extremely sparse cover of vascular plants. Therefore, it is not surprising that no sawfly species are known from this biome (Figs 21, 22).

**Fig. 21.** The Namib Desert Biome in the vicinity of the Gobabeb Research Station (eastern Namibia). (Photo by J. Deckert)

**Fig. 22.** The Namib Desert Biome, north of Swakopmund (Namibia). (Photo by F. Koch)

## 3. Habitats of Sawflies

Profound statements on habitat preference are really only possible where the abundance of species is also relatively high, such as in many ecosystems of the Palaearctic Region.

In comparison, however, the abundance of Symphyta in southern Africa is significantly lower, so it is much more difficult to reach corresponding conclusions. Based on previous field experiences, observations on habitat preference were most readily obtainable in the Afromontane region in north-eastern South Africa, the Drakensberg Mountains (Koch 2005a, b).

Within the area treated here, appropriate investigations were only possible in the Woodland Savanna Biome, especially in the riverine vegetation associated with the Kunene and Okavango rivers (Fig. 23). In the Thornbush Savanna Biome it was possible to make appropriate observations in the Waterberg area in Namibia. Very good habitats favoured by sawflies are located at the foot of the Waterberg, where the humidity is at an optimum, resulting in the presence of luscious tree

**Fig. 23.** Riverine vegetation at the Okavango River in the vicinity of Popa Falls (West Caprivi, northern Namibia) (Photo by M. Uhlig)

**Fig. 24.** Dense vegetation of herbaceaous plants in the semi-shade at the foot of the Waterberg in Namibia. *Athalia incomta*, *A. marginipennis*, *A. ustipennis*, *Xenapates similis* and *Distega montium* occur in this habitat. (Photo by F. Koch)

and herbaceous vegetation. Sawflies were observed to have a high preference for partially shaded areas in this habitat (Fig. 24).

Based on these experiences from the Waterberg we propose that in the other biomes where sawflies are inconspicuous and difficult to find (Nama Karoo Biome, Succulent Karoo Biome and Fynbos Biome), that Malaise traps are erected in the semi-shade to maximise returns.

## 4. The history of the study of the sawflies of the Afrotropical Region

The study of the Symphyta of the Afrotropical Region commenced about 200 years ago. The first species, *Arge capensis* (Klug, 1814) was described as *Hylotoma capensis* and collected at the Cape of Good Hope. Twenty years passed before the description of five other *Hylotoma* species from the former Cape Province (South Africa) and three species from West Africa (Klug 1834) (Fig. 25A). In the same year the description of *Athalia himantopus* Klug, 1834 from the Cape Province was published, the first record of an Afrotropical tenthredinid.

Almost 50 years later, additional species were described by Gribodo (1879) (Fig. 25B), W.F. Kirby (1882) (Fig. 25C) and Buysson (1898) (Fig. 25D). With the steady

increase of extensive research trips to Africa and the establishment of mission stations at the beginning of the 20th century, the number of new taxa increased significantly (Konow 1904, 1907a, b, c, d, 1908, Mocsáry 1909, Enslin 1911, 1912, 1913a, b, Enderlein 1919, 1920) (Figs 25E-H).

This increase in the number of newly described genera and species led to confusion that needed to be resolved in first taxonomic revisions. In this respect, the identification keys by Konow (1908) for the genus *Athalia* Leach and Enslin (1913a)


**Fig. 25. A-K**. A selection of important entomologists who contributed considerably to the knowledge of Afrotropical sawflies. **A**. J.C.F. Klug (1775-1856). **B**. G. Gribodo (1846-1924). **C**. W.F. Kirby (1844-1912). **D**. H. du Buysson (1856-1927). **E**. F.W. Konow (1842-1908). **F**. A. Mocsáry (1841-1915). **G**. E. Enslin (1879-1970). **H**. G. Enderlein (1872-1968). **I**. R. Forsius (1884-1935). **J**. J.-J. Pasteels (= J. Pasteels) (1906-1991). **K**. R.B. Benson (1904-1967). (Portrait collection of the SDEI)

for the genera *Xenapates* Kirby, *Neacidiophora* Enslin and *Dulophanes* Konow, *Trisodontophyes* Enslin and *Distega* Konow were of particular value. Besides a few descriptions in the following 15 years, the knowledge of the Afrotropical Symphyta only received a new input by Forsius (1927a, b, c, 1928a, b, c, 1930a, b, 1931) (Fig. 25I), however, he did not revise the existing identification keys.

Finally, Pasteels (1949, 1950, 1951, 1952, 1953a, b, c, 1954a, b, 1955a, b, c, 1963) (Fig. 25J) who dealt mainly with the fauna of the Belgian Congo [Democratic Republic of the Congo] and the neighboring eastern provinces, described several new species, and provided urgently needed determination keys. Particularly noteworthy in this context are the revisions of the genera of the families Argidae (Pasteels1953a, 1955b, 1963) and Tenthredinidae (Pasteels 1949, 1955c) with the exception of the genus *Athalia*.

Since Konow (1908) the species-rich genus *Athalia* was scarcely worked on and was revised only by Benson (1962) (Fig. 25K) within the framework of the revision of the world Athaliini. Over the next 30 years, only a few *Athalia* species were sporadically described (Viitasaari & Kontuniemi 1976, Muche 1981).

In 1992, with the beginning of the Entomological Expeditions of the MFN, studies on sawflies in the Afrotropical Region, especially in southern Africa, received new scientific interest. This era saw the commencement of targeted searching for sawflies in all biomes. Resultant data demonstrated that the Afromontane region is particularly rich both in species and abundance. In addition the large reference

**Fig. 26.** An impression of the entomological collection of the Agricultural Research Council, Plant Protection Research Institute, Pretoria, South Africa with its curator Dr. Connal Eardley. (Photo by C. Eardley)

**Fig. 27.** An impression of the entomological collection of the Iziko South African Museum, Cape Town, South Africa with its curator Dr. Simon van Noort. (Photo by Aisha Mayekiso)

**Fig. 28.** An impression of the entomological collection of the IITA, Cotonou, Benin. (Photo by F. Koch)

collections in southern Africa (AMGS, NMNW, PPRI (Fig. 26), SAMC (Fig. 27), TMSA) and the collection of IITAC (Fig. 28) in Benin (West Africa) were evaluated. Unfortunately, the entomological collections in Kenya (ICIPE, NMKE) are not readily available for examination.

In Europe the evaluated material from Africa is scattered over various institutions including the larger (BMNH, RBINS, MNHN, MRAC,) and smaller collections (MFN, SDEI, UZMT, MCSN, ZSM). Large museums in the United States are becoming increasingly important for African studies (CASC, LACM, SEMC, USNM).

In the wake of this taxonomic research, revisions of several large genera and species groups were published: *Xenapates* (Koch 1995), *Neacidiophora* (Koch 1998a, b), *Trisodontophyes* (2001), *Triarge* (Koch 2006b), *Durbadnus* (Koch & Liston 2012c), *Athalia vollenhoveni* species group (Koch 2006), *Athalia himantopus* species group (Koch 2007), *Arge mirabilipes* species group (Koch & Eardley 2011), *Arge capensis* species group (Koch & Liston, 2012a) and *Xenapates variator* species group (Koch 2012b).

As a result of these taxonomic-systematic studies, the known number of species was increased considerably. For example Pasteels' (1949) revision of *Xenapates* treated 17 species, while currently 46 valid species are recognised. In *Neacidiophora* the number of species has correspondingly increased from 10 to 15, in *Trisodontophyes* from 7 to 20 and in *Triarge* from only one species (Pasteels 1953a) to 9 species.

The situation is similar in the *Athalia vollenhoveni* species group for which Benson (1962) mentioned 6 species or for the *A. himantopus* species group for which Benson (1962) recorded only one species with two other subspecies. The revisions of these species groups by Koch (2006c, 2007) documented 10 and 8 valid species respectively.

Reassessment of species distribution has demonstrated that the ranges of several species are smaller than described by Benson (1962). According to Benson (1962) for instance *A. vollenhoveni* is known from Ethiopia, Kenya, Uganda and Tanzania [Tanganyika]. However, this species is distributed only in Ethiopia and in the Arabian Peninsula (Yemen). The artificially large range was a result of misidentifications of specimens attributed to this species.

These taxonomic studies and associated intensive field work, especially in Namibia and South Africa, significantly expanded the species lists for these countries. For example in 1887, A.W. Eriksson collected the first Namibian specimens from the Okavango River (Koch 2001). Since then the Namibian Symphyta have been poorly studied, with only two species being recorded prior to the commencement of this survey in 1992, namely: *Athalia turneri* Forsius (Tenthredinidae) and *Arge stuhlmanni* (Kohl) (Argidae) (Pasteels 1953a). Possibly, the low abundance and poor diversity may be due to the arid environment with xerophytic associated

vegetation, but is probably also a result of a general lack of scientific interest in the group.

Based on the experience gained during field work and after evaluation of various reference collections Koch (2000) listed 16 species for Namibia. In this monograph 26 species are recorded from Namibia.

South Africa is biogeographically more diverse than Namibia, especially the Afromontane Region in the east, and with its high diversity of plant species a more species rich sawfly fauna is to be expected. Following intensive field work in the Drakensberg mountain system a total of 51 species for South Africa were recorded (Koch, 2005b). Further research focused on the South African fauna elevated the recorded total to 82 species.

Hitherto, 11 species were known from the South African part of the study area, including three introduced species. As a result of further intensive investigations in this area, a total of 29 species are now documented.

## 5. Material and methods

### 5.1 Collecting methods

In southern Africa three collecting methods have proven to be successful for sawfly collecting. Assessment of field data illustrated that the best returns are gained by combining all three collecting methods.

#### Malaise trap

This trap is named after the Swedish entomologist René Malaise (1892-1978) (Fig. 29), who developed this trap for insect collecting in South East Asia. He designed the trap based on his experiences of observing the behavior of insects in a normal living tent. He modified the tent design to exclude the side walls, but with a middle

**Fig. 29.** René Malaise (1892-1978), a Swedish entomologist. He was one of the most important specialists of Symphyta of the world, and developed the insect trap which is named after him.

wall and one higher middle corner which is connected to a killing bottle. The fore and back sides as well as the roof are made of white netting, whereas the middle wall is made of black netting, but other combinations of colour are possible (Fig. 30). The insects fly into the middle wall and then make their way to the highest corner of the trap which opens into a collecting bottle, containing an insecticide to kill the insects and keep them dry. Additionally, the bottle contains loosely crumbled soft paper to separate the dying insects. This has the advantage that the insects do not destroy each other. A commonly used alternative is to charge the collecting head with 80-96% ethanol which kills and preserves the specimens making them suitable for DNA extraction.

The collecting bottle should be serviced daily for dry collections and, if possible, in the early morning to verify whether no new insects have just been trapped in it. If serviced later, a few drops of ethyl acetate must be added to speed up the killing of insects. If ethanol is used in the collecting head, the traps can be serviced at longer intervals, two weeks or more. The Malaise trap should be positioned in a flight path between dense vegetation present on at least the front side or preferably on both sides to facilitate the channelling of insects into the trap. Also best results it the trap head is pointed to the morning sun.

**Fig. 30.** Malaise trap in the Marloth Nature Reserve (Western Cape Province) with yellow pan trap in the foreground. (Photo by F. Koch)

Since 1992 the number of Malaise traps has continuously been increased so that from 1997 on field work has regularly been conducted with 25 traps. The field work was carried out each time over a period of approximately six to eight weeks, mostly during or after the rainy season, from November to March in the summer rainfall zone, and from August to November in the winter rainfall zone. Several National Parks in Namibia and South Africa were visited, each for a period of approximately 5 to 8 days. Advantages of Malaise traps are that they are up continuously, 24 hr/day, and trap many insects from low lying and dense vegetation that are difficult to get by hand collecting.

In private areas, especially in South Africa, it was possible, with the consent of the landowners, to leave the traps running over a longer period (almost two months). The landowner assisted with servicing the traps on a weekly basis.

**Coloured pan traps (yellow pan traps)**

Pan traps are easy to use and transport, but they produce wet sawflies. A pan trap is a shallow dish filled with soapy water. Usually only several drops of soap are necessary to reduce surface tension. The soap reduces the surface tension of the water and prevents the sawflies from escaping when they drift to the side. Foam on the water surface should be avoided.

Yellow coloured pans are most common (Fig. 30), but white and blue pans are used as well since they attract different species. For sawflies, especially *Athalia* species, yellow pan traps are most effective in northern Germany and southern Britain (Ritzau 1988, Barker *et al.* 1997).

Between 1981 and 1983 in Burundi more than 4000 sawfly specimens (95 % *Athalia*) of about 20 species were collected by means of yellow pan traps (Chevin 1985). These results have been confirmed in southern Africa, and additionally yellow pans have been shown to be highly effective for collecting *Xenapates similis*.

We observed that the use of different yellow tones attracts sawflies in different ways. An intense yellow colour is most effective to trap sawflies.

Since there is no preservative in the soapy water, traps used in hot regions need to be checked within four-hour intervals, so that the muscle tissue of the sawflies does not decompose. Sawflies collected in this way should be subsequently preserved in 70-96% ethanol after having been thoroughly rinsed to remove any soap residue.

An alternative is to use propylene glycol as the preservative in the pans. This allows for a longer interval between servicing since this inert chemical is highly viscous and does not evaporate. It is also a hydrophilic chemical rapidly dehydrating specimens and thereby preserving DNA for molecular analyses. Specimens can be transferred directly from the collecting fluid (propylene glycol) and preserved in 96% ethanol. Propylene glycol is environmentally safe and non-toxic to vertebrates,

which sometimes drink the contents of pan traps. The very similar monoethylene glycol (used as antifreeze in vehicle radiators) is highly toxic to vertebrates and should not be used.

### Hand nets

Hand nets are good tools for sawfly collecting as they allow specimen acquisition without too much damage, and make ecological observation possible (Fig. 31). A hand net consists of a handle and a circular frame supporting a gauze bag at the

**Fig. 31.** Entomologist in the field, using a hand net and observing insects, in the habitat of *Caliroa blanki*, Lekgalameetse Nature Reserve, Limpopo Province. (Photo by F. Koch)

end. It is an advantage if the handle is constructed from an aluminum telescoping pole allowing for extension to reach further into inaccessible vegetation. The unextended handle should be about one meter long. The circular frame should be constructed from strong, but lightweight flat material, with a diameter of 30 to 35 cm and be firmly attached to the handle, but should still be detachable so that the netting bag can be easily replaced. The netting bag should be about 60 cm long, made of a fine, light, yet strong netting with a mesh width of about 1 mm. White material makes it easier to see the sawfly in the net.

Specimens collected by hand net should subsequently be transferred to a small plastic bottle with some loosely crumpled soft paper and a few drops of ethyl acetate for killing. Specimens collected by hand net destined for further molecular biological analyses should be transferred directly into small tubes containing ethanol (≥96%) to kill and preserve the specimens.

## 5.2 Transport of dry insects in the field and further preparation

Samples collected using Malaise traps are very rich both in number of specimens and species of various orders of insects mainly Hymenoptera, Diptera, Coleoptera and Lepidoptera. If the specimens are killed in the collecting head with insecticide then this material should first be sorted and cleaned *i.e.*, remove moth wing scales. The selected insects are then placed on thin wadding sheets and enclosed in paper envelopes (Fig. 32). These envelopes can be stacked in a stable cardboard

**Fig. 32.** Worktable during the field work with cardboard box and the material collected using Malaise traps and selection of insect groups. (Photo by F. Koch)

box (no plastic - risk of mold) and transported safely. If ethanol or propylene glycol is used in the collecting head, the specimens can be transferred to a bottle with the collecting fluid.

In the laboratory, the envelopes should be carefully placed in a humidifying chamber, and after a few hours the insect material will be soft enough for preparation. The collected sawflies should be pinned using stainless steel insect pins, usually of size N°0 and N°1 which should be inserted through the right lateral lobe of the mesoscutum.

Subsequently, the specimens need to be labeled with details of the collecting location: country, detailed locality, geographical coordinates, altitude, date and collector, and including collecting method and habitat details. It is common for material from the Afrotropical Region to be designated using blue labels, but this colour assignment varies across different institutions (Fig. 33).

For further scientific study the sawflies need to be pinned in an insect drawer and as a further measure against potential pest infestation deposited in the freezer for about one week before integration into the main collection (Fig. 34).


**Fig. 33.** A sawfly (*Distega montium*, female) mounted on a continental pin, specimen positioned high enough to allow attachment of locality, determination and other labels as well as a plastic capsule containing dissected ovipositor.
(Photo by H. Goetz)

**Fig. 34.** Use of unit trays in a drawer. Curated (pinned and labelled) sawfly material after field work and sorting of samples.
(Photo by H. Goetz)

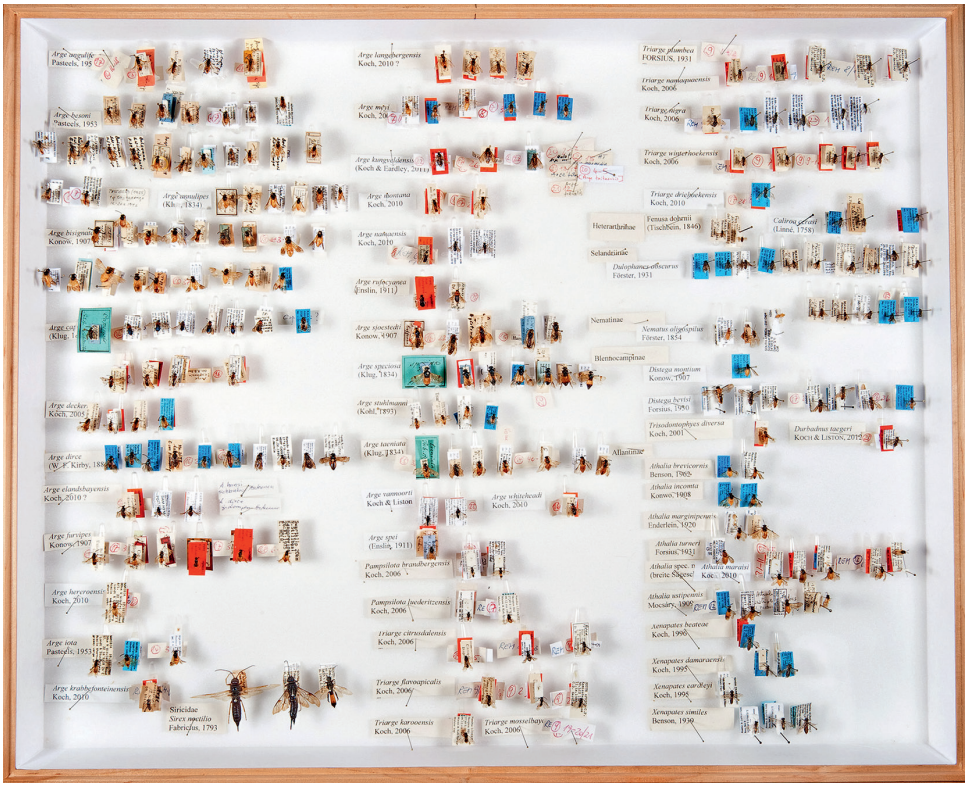

**Fig. 35.** Drawer containing representatives of all species studied during preparation of this book, including numerous type specimens (with red labels). (Photo by H. Goetz)

Permanent long term storage and conservation of the specimens depends on the individual and institutional custodial considerations and scientific requirements, but the specimens should be housed in light- and dust-proof drawers contained in a sealed room with environmental control facilities to regulate low humidity and temperature to reduce potential damage by pests. Fumigation and visual inspection of the drawers on a regular basis is essential to identify and contain potential pest infestation (Fig. 35).

## 5.3 Preparation of genitalia

For reliable identification, it is necessary to study the genitalia of most sawfly species (**Chapter 8.1: Morphology of adults**).

Specimens are softened in a moist chamber, and depending on the age of the material and the body size the softening process takes varying lengths of time. Large and very old material should be soft after about 24 hours. For fresher material

and smaller species a few hours is usually sufficient. However it is preferential to dissect the genitalia of the specimens immediately after pinning while they are still fresh.

The dissected parts of the genitalia including the glued parts of the copulatory organs should be stored in colourless PCR tubes, 0.2 ml, which are mounted on the pin below the specimen (Fig. 33). It is also possible to carefully mount the glued parts of genitalia separately on a card also mounted on the pin.

**Genitalia of males**

The dissection of the male genitalia is performed in dorsal view under a stereoscopic-microscope. The abdomen of the specimen is held with tweezers and with an insect pin the genital capsule is easily completely extracted and transferred into 80% ethanol. Further dissection of the genitalia is performed (removal of the penis valve and / or of the digitus, cuspis and parapenis as well as harpe) in ethanol. With two sharp insect pins the attached muscle fibers are carefully removed to expose the sclerotized structures. After cleaning, the penis valve is placed in 96% ethanol for a few minutes and then transferred into xylene for a short time for dehydration.

In the meantime, the remaining parts of the genital capsule should be glued on a small plastic card. If necessary the digitus and cuspis can be prepared for scanning electron microscope investigation at this stage. A transparent plastic sheet with a size of 10 mm x 3 mm is cut out and used as a small microscope slide and prepared with a drop of Canada balsam. The penis valve should now be accurately embedded into the Canada balsam. Finally, the drop of balsam containing the penis valve should be carefully covered with a small cover slip (3.0 mm x 3.0 mm). This preparation allows for examination of the genitalia with a transmission-light microscope.

**Genitalia of females**

The dissection of the lancet is also performed under a stereoscopic-microscope, however this is done in ventral view. The abdomen is held with tweezers and with an insect pin the entire ovipositor is pulled out of the sawsheath. Can also be held between thumb and fore finger and then the ovipositor pulled down with a pin. Attempts should be made to separate the left and right half of the ovipositor. If possible, the lancet is separated from the lance, and then cut with a sharp razor blade at the base, so as to leave as many parts as possible of the genitalia in the specimen. Subsequently the lancet is transferred for cleaning into 80% ethanol. Further treatment is carried out as stipulated above for the penis valve of the male.

The preparation of the hypopygium is carried out with a very sharp pin, which is used to separate the apical sternum basally and laterally. This process is often very difficult for old material. Subsequently the dissected hypopygium is carefully freed from attached muscle fibers and cleaned in 80% ethanol and then smoothed and glued on a small plastic card.

### 5.4 Barcoding analysis

Barcoding analysis (Blank *et al*. 2013) was conducted for the following species for which suitable material was available: *Athalia incomta* and *A. ustipennis*.

DNA extraction was attempted for a considerable number of additional specimens and species, but conventionally collected material was often too old or had been treated in an unsuitable way (*e.g.*, specimens re-moistened for mounting subsequent to collection). For DNA extraction the single leg of an imago was removed and submitted to the Canadian Centre for DNA Barcoding (CCDB) in Guelph, Canada, where the DNA sequencing was performed. The DNA extracts are stored at the CCDB, the vouchers at the SDEI.

DNA extraction, PCR amplification, and sequencing were conducted using standardised high-throughput protocols (Ivanova *et al*. 2006, DeWaard *et al*. 2008). The target region has a length of 658 bp, starting from the 5' end of the mitochondrial cytochrome c oxidase I (COI) gene and includes the 648 bp barcode region used as standard in the animal kingdom (Hebert *et al*. 2003). Sequence data can be obtained through BOLD (http://www.barcodinglife.com/) and include LIMS report, primer information, and access to trace files. Sequences were aligned using the BOLD Aligner.

Sequence divergence statistics were calculated using the Kimura 2-parameter model. Genetic distances were calculated using analytical tools in BOLD and are given as maximum pairwise distances for intraspecific variation and as minimum pairwise distances for interspecific variation. Specimens without a Binary Index Number (BIN) were excluded from these calculations but some with shorter sequences were subsequently included to associate imagines. Clusters of similar sequences are denoted by a Globally Unique Identifier (GUID) that is registered in BOLD.

## 5.5 Abbreviations used in the text

BIOTA : Biodiversity Monitoring Transect Analysis in Africa.
BMBF : Federal Ministry of Education and Research, Germany.
DFG : German Research Foundation, Germany.
NRF : National Research Foundation, South Africa.

Material examined originated from the following institutions:

AMGS : Albany Museum, Grahamstown, South Africa.
BMNH : The Natural History Museum [formerly British Museum (Natural History)], London, UK.
CASC : California Academy of Sciences, San Francisco, USA.
DEUS : Department of Entomology, University of Stellenbosch, South Africa.
DMSA : Natural Science Museum, Durban, South Africa.

HNHM : Hungarian Natural History Museum, Budapest, Hungary.
ICIPE : International Centre of Insect Phylogeny and Ecology, Nairobi, Kenya.
IITAC : International Institute of Tropical Agriculture, Cotonou, Benin.
LACM : Los Angeles County Museum of Natural History, Los Angeles, USA.
MCSN : Museo Civico di Storia Naturale 'Giacomo Doria', Genoa, Italy.
MFN : Museum für Naturkunde Berlin, Germany.
MNHN : Muséum d'Histoire Naturelle, Paris, France.
MRAC : Musée Royal de l'Afrique Centrale, Tervuren, Belgium.
NHMW : Naturhistorisches Museum, Wien, Austria.
NHRS : Naturhistoriska Riksmuseet, Stockholm, Sweden.
NMBZ : Zimbabwe National Museum, Bulawayo, Zimbabwe.
NMKE : National Museum of Kenya, Nairobi, Kenya.
NMSA : Natal Museum, Pietermaritzburg, South Africa.
NNIC : Namibian National Insect Collection, Windhoek, Namibia.
OLML : Oberösterreichisches Landesmuseum, Linz, Austria.
PPRI : ARC-Plant Protection Research Institute, Pretoria, South Africa.
RBINS : Royal Belgian Institute of Natural Sciences Brussels, Belgium.
RMNH : Nationaal Natuurhistorische Museum (Naturalis), Leiden, Netherlands.
SAMC : Iziko South African Museum, Cape Town, South Africa.
SDEI : Senckenberg Deutsches Entomologisches Institut, Müncheberg, Germany.
SEMC : Snow Entomological Museum, University of Kansas, Lawrence, Kansas, USA.
SMNS : Staatliches Museum für Naturkunde, Stuttgart, Germany.
TMSA : Ditsong National Museum of Natural History (formerly Transvaal Museum), Pretoria, South Africa.
USNM : National Museum of Natural History, Smithsonian Institution, Washington, D.C., USA.
UZMT : Zoological Museum, University of Turku, Finland.
ZMPA : Museum of the Institute of Zoology, Polish Academy of Science, Warszawa, Poland.
ZMUC : Zoological Museum, University of Copenhagen, Copenhagen, Denmark.
ZMUH : Zoologisches Institut und Zoologisches Museum, Universität Hamburg, Hamburg, Germany.
ZSM : Zoologische Staatssammlung, Munich, Germany.

**The following measurements are commonly used (Figs 43B, C)**

POL : Postocellar line; the distance between the mesal margins of the two lateral ocelli (Fig. 43C).
OOL : Ocular ocellar line: the distance between a compound eye and a lateral ocellus (Fig. 43C).
MS : Malar space; the shortest distance between the base of the mandible and the edge of the compound eye (Fig. 43B).
IA : Interantennal area; the shortest distance between the inner margins of the toruli (Fig. 43B).

LC : Length of the clypeus; the vertical distance between the epistomal suture and the anterior margin of the clypeus medially (Fig. 43B).

**The following ratios are applied:**

POL : OOL
MS : IA
IA : LC

## 6. Life cycle

Very few observations exist about mating, oviposition and preimaginal stages of Afrotropical sawflies. Essentially, their life cycle does not differ fundamentally from the partly well-studied cycles of sawflies of other biogeographical regions (Viitassari 2002). Hence the following description of the life cycle of Tenthredinoidea is described in generalized terms but pertaining to Afrotropical species whenever possible.

After mating (Fig. 36) the females use their saw-like ovipositor to cut into the host plant tissue for egg laying (Fig. 37A), usually on the edge of a leaf. Also leaf surface, along midrib or main veins, in twigs, stems, buds. The eggs are mostly laid singly and are visible below the epidermis (Fig. 37B). The incubation period of the eggs is generally rather short and is dependent on ambient temperature and

**Fig. 36.** *Athalia ustipennis* Mocsáry during copulation. (Photo by J. Deckert)

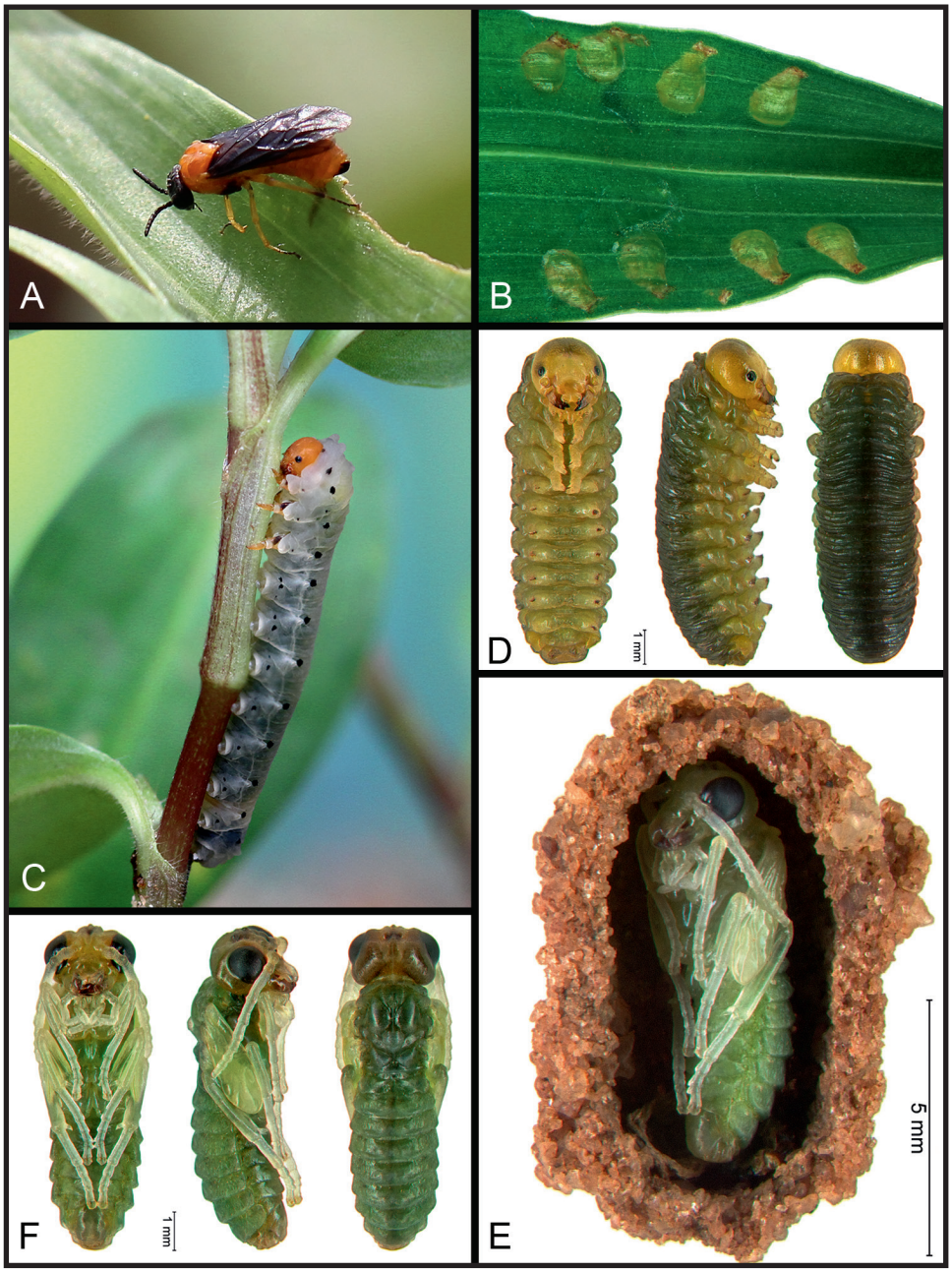


**Fig. 37. A-F**. **A**. *Distega nigeriae* Forsius during egg laying (ovoposition). The ovipositor is visible in the plant tissue as a darker shadow. **B**. Eggs of *D. nigeriae* in a leaf of *Commelina benghalensis*. **C**. The larva of *D. nigeriae*. **D**. The prepupa of *Xenapates gaullei*, ventral, lateral and dorsal (from left). **E**. The opened cocoon containing pupa of *X. braunsi*. **F**. The pupa of *X. braunsi*, ventral, lateral and dorsal (from left). (Photo by G. Goergen)

humidity. Usually the egg stage encompasses one to four weeks with apparent variability between species.

After hatching the first instar larva immediately begins to feed externally on plant foliage or internally for *Fenusa dohrnii* and *Sirex noctilio*.

As the larvae grow, they molt several times (Fig. 37C). The larval developmental phase includes five or six instars and takes between three and six weeks, depending on environmental conditions.

During this time the larvae are susceptible to predation, especially predatory beetles, flies, bugs, wasps and ants. In addition birds and small mammals eat the larvae.

When the last instar is completed, a non-feeding stage called the prepupa (Fig. 37D), searches for a suitable pupation site. This site can be in the soil, in stems or rotten wood, or among other plant material.

Some prepupae spin cocoons and others form a cell in the soil or other substrate (Fig. 37E). Under temperate climatical conditions the prepupa of most sawflies overwinters, and goes into diapause until the following spring. The species that have overwintered as prepupa pupate during the early spring. The duration of the pupal stage (Fig. 37F) is short and adults generally emerge after two or three weeks. Adult sawflies usually live for only several days up to a week or two.

Little is known of the phenology of the tropical sawfly species (Smith 1995). However, almost all species and individuals occur during the rainy season (Smith & Janzen 2003a, 2003b, Smith & Grisell 2014).

Under subtropical and tropical climatic conditions with dry and rainy seasons it is fairly certain that diapause occurs in the dry season, when the host plants are not present. This is especially true for the winter rainfall zone of the study area. Therefore one generation (monovoltism) of the species of this area seems to be obligatory. Smith (1995) confirmed these observations for the seasonal dry forests of Costa Rica, where the adults of several species of Tenthredinidae and Argidae can be common shortly after the commencement of the wet season, when trees are flush with new growth.

In wetter areas some sawfly species have several generations annually (Smith 1995). This result has been confirmed for *Athalia marginipennis* Enderlein in Burundi (Koch 2007), when the flight season extends over several months. This extended flight season in turn provides evidence for the existence of at least two (bivoltine) or more generations annually (polyvoltine). An obligatory diapause is probably not present or very short between these generations.

This limited knowledge underlines the need to thoroughly research the ecology and phenology of the Afrotropical Symphyta.

## 7. Host plants

The host plant relationships of Afrotropical sawflies is very poorly known when compared to the Palaearctic Region (**Chapter 1: Introduction**). The few known associations in literature are questionable since the identification of some species has not been unambiguously clarified.

In this respect, the present monograph is of value because the validity of the species treated here has been thoroughly checked. Additionally, for most species data are available based on observations about their habitat or preferences for certain plant species. The included data provide a valuable baseline, especially for local entomologists and ecologists so as to continue further investigations into this still largely unexplored topic of host plant relationships (**Chapter 1: Introduction**).

The following information on host plant relationships for Afrotropical species exists in the literature:
Sorauer (1928): ***Athalia sjoestedti*** Konow, 1907 and ***A. flacca*** Konow, 1907: Turnips in Kenya, Tanzania and Zimbabwe.

Saraiva (1939): ***Athalia flacca*** Konow, 1907 (taxonomic status not verified): Turnips, cabbage and other cruciferous plants (Brassicaceae).

Lepelley (1959): ***Athalia himantopus*** Klug, 1834 (not known from Kenya and Uganda, probably *A. marginipennis* Enderlein, 1920): *Brassica oleracea* Linnaeus in Kenya and on *B. rapa* Linnaeus in Uganda; ***Athalia segregis*** Konow, 1907: Crucifers (Brassicaceae) in Kenya; ***Athalia sjoestedti*** Konow, 1907: *Brassica oleracea* Linnaeus in Tanzania and on *B. rapa* Linnaeus in Kenya and Tanzania, on crucifers (Brassicaceae); *Zea mays* Linnaeus (Poaceae) in Tanzania; ***Athalia vollenhoveni*** Gribodo, 1879 (probably *A. sjoestedti* Konow, 1907): Crucifers (Brassicaceae) in Kenya; ***Neacidiophora athaloides*** (Konow, 1907) on *Cissus adenocaulis* Steudel ex A. Richard (Vitaceae) in Kenya and Uganda.

Nonveiller (1984): ***Athalia schweinfurthi atripennis*** Benson, 1962 [= *A. atripennis* Benson, 1962]: *Brassica napus* Linnaeus and *Raphanus sativus* Linnaeus (Brassicaceae) in Cameroon.

According to Benson (1962) in the revision of the Athaliini: ***Athalia furvipennis*** Konow, 1907 (taxonomic status not verified): larvae on various Brassicaceae, pest on *Brassica rapa* Linnaeus; ***Athalia sjoestedti*** Konow, 1907: larvae on various Brassicaceae, pest on *Brassica rapa* Linnaeus and *B. oleracea* Linnaeus; ***Athalia himantopus*** Klug, 1834: larvae on *B. oleracea* (Brassicaceae); ***Athalia himantopus truncata*** Enslin, 1913 [= *A. truncate* Enslin, 1913]: *Nasturtium* spp. (Brassicaceae); ***Athalia vollenhoveni*** Gribodo, 1879: larvae on various Brassicaceae, pest on *Brassica oleracea* Linnaeus (Brassicaceae). ***Athalia schweinfurthi*** Konow, 1891 [= *A. vollenhoveni* Gribodo, 1879]: *Lepidium sativum*

Linnaeus (Brassicaceae); ***Athalia mellis*** Benson, 1962 [ = *A. mashonensis* Enslin, 1911]: *Coeus barbutus* Bentham = *Plectranthus barbatus* Andrews (Lamiaceae); *Salvia* sp. (Lamiaceae).

Based on chemical analyses derived from the substances of host plants that are sequestered in the adults of reliably identified species of *Athalia*, Opitz *et al*. (2012) associated host plants of the order Lamiales and the family Brassicaceae with these species:
*Athalia excisa* Koch, 2006 (Brassicaceae)
*Athalia flavobasalis* Koch, 2007 (Brassicaceae)
*Athalia guillarmodi* Benson, 1956 (Brassicaceae)
*Athalia himantopus* Klug, 1834 (Brassicaceae)
*Athalia incomta* Konow, 1908 (Lamiales: Scrophulariaceae)
*Athalia marginipennis* Enderlein, 1920 (Brassicaceae)
*Athalia obsoleta* Benson, 1962 (Brassicaceae)
*Athalia ustipennis* Mocsáry, 1909 (Brassicaceae)
*Athalia vollenhoveni* Gribodo, 1879 (Brassicaceae)

Previously nothing was known about the host plants of *Xenapates* W.F. Kirby, 1882 species. However, in the context of cooperation between the authors and breeding experiments in the laboratory (IITA, Benin) host plant relationships for four West African species has been established.

Larvae of the widespread *Xenapates braunsi* (Konow, 1896) were found on following Poaceae: *Digitaria horizontalis* Willdenow (Jamaican crabgrass), *Pennisetum purpureum* Schumacher (elephant grass) and *Setaria barbata* ( Lamarck) Kunth (bristly foxtail grass) (Poaceae), as well as *Zea mays* Linnaeus (corn, maize) (Poaceae). Larvae of another common species, *Xenapates gaullei* (Konow, 1896), were observed on *Commelina communis* Linnaeus (Asiatic dayflower) and *C. benghalensis* Linnaeus (Bengal dayflower) (Commelinaceae) (Liston *et al*. 2015).

All four of the larval hosts of *X. braunsi* identified in this study are of greater or lesser importance throughout tropical and sub-tropical Africa as cereal or fodder crops. The host plants of *X. gaullei* (*Commelina* spp.) are also of direct interest to man. Leaves of *C. benghalensis* are eaten as a vegetable in Africa and parts of Asia, whereas *C. communis* is better known internationally as a troublesome invasive weed, for example in parts of Europe and North America. Both *Xenapates* species could therefore be regarded as potential crop pests.

Furthermore, the larvae (Fig. 37C) of the West African *Distega nigeriae* Forsius, 1927b were observed feeding on *Commelina benghalensis* and *C. communis* as well as *Digitaria horizontalis* in Benin including recording of the complete metamorphosis (G. Goergen, unpublished). *Distega nigeriae* Forsius, 1927 **sp. rev.** is considered to be a valid species, and not a synonym of the East African *D. mocsaryi* Enslin, 1913b as treated by Pasteels (1955). The holotype of *D. nigeriae* was compared with the lectotype of *D. mocsaryi* (Koch in prep.). *Distega*

*nigeriae* has a nearly entirely yellow mesonotum, which is predominantly black in *D. mocsaryi*. Furthermore, the serrulae of *D. nigeriae* are more slender than those of *D. mocsaryi*. A more detailed explanation will be presented in a separate revision (Koch in prep.).

Adults commonly visit flowers or leaves of plants other than the larval hosts (Smith 1989). For example Benson (1962) mentioned adults of *Athalia schweinfurthi* Benson, 1962 on flower heads of *Senecio elgonensis* T. Fries (*Dentrosenecio elgonensis* E.B. Knox) (Asteraceae). This observation was confirmed during many hours of field work in southern Africa. For example in the Lekgalameetse Nature Reserve, Limpopo Province, South Africa *Athalia gessi* Koch, 2003 and *A. mashonensis* Enslin, 1911 were sampled in large numbers on the flowers of the Lemon Bush / Fever tea, *Lippia javanica* (Burman f.) Sprengel (Verbenaceae) (Koch 2003).

A further example is *Arge deckerti* Koch, 2005, that is observed in large numbers on flowers of *Cyphostemma congestum* (Baker) B.M. Descoings ex. Wild & R.B. Drumm (Vitaceae) (cover photo, Fig. 60) and *Nidorella resedifolia* A.P. de Candolle (Asteraceae) (Fig. 61). In Ndumu Game Reserve (KwaZulu-Natal, South Africa) males were observed feeding on pollen in the flowers of *Nidorella auriculata* A.P. de Candolle (Asteraceae).

In the eastern provinces of South Africa and Zimbabwe *Athalia incomta* Konow, 1908 is sampled regularly in large numbers on leaves of the widely cultivated ornamental plant Cape Honeysuckle, *Tecomaria capensis* (Thunberg) Spach (Bignoniaceae), which is probably not the host plant. In the mountain regions of the Limpopo and Mpumalanga provinces adults were collected on *Helichrysum krausii* (Schultz Bipontinus) (Asteraceae). In the study area this species was observed on *Selago dinteri* Rolfe (Scrophulariaceae, Lamiales), which could be the host plant (*vide* Opitz *et al*. 2012).

At the foot of the Waterberg (Namibia), adults of *Athalia ustipennis* Mocsáry, 1909 were repeatedly observed on the shrubby tree *Grewia flavescens* Jussieu (Tiliaceae). Adults of *Caliroa blanki* Koch & Smith (2011) were also very numerous on leaves of the shrub *Bauhinia galpinii* N.E. Brown (Fabaceae) in Lekgalameetse Nature Reserve (Limpopo Province, South Africa).

Nothing is known about the host plants of the species of *Xenapates* which occur in the study area, with the exception of *Xenapates beateae* Koch,1996 and *X. damaraensis* Koch, 1995. Adults of these two species were observed on leaves of *Achyranthes aspera* Linnaeus var. *sicula* Linnaeus (Amaranthaceae) in a small isolated moist habitat in the Kaokoveld of Namibia (Fig. 141).

Data on detailed host-plant associations for various species are provided in the special taxonomic section (**Chapter 9: Systematic presentation**) to facilitate future research investigation.

Nothing is known about host-plant relationships of the species in the genera *Pampsilota* Konow, 1899, *Triarge* Forsius, 1931, *Trisodontophyes* Enslin, 1911, *Durbadnus* Pasteels, 1954 and *Dulophanes* Konow, 1907d. The species of *Dulophanes* as Selandriinae, feed possibly on fern or mosses.

## 7.1 The host plants of the species of the study area

For introduced (aliens) and invasive species:
*Nematus oligospilus*: Different willows (*Salix babylonica* Linnaeus, *S. fragilis* Linnaeus (Salicaceae) (Urban & Eardley 1995, 1997, Koch & Smith 2000).
*Fenusa dohrnii*: *Alnus* spp. (alder) (Betulaceae).
*Caliroa cerasi*: *Prunus persica* (Linnaeus) Batsch (peach) (Rosaceae).
*Prunus armeniaca* Linnaeus (apricot) (Rosaceae).
*Prunus domestica* Linnaeus (plum) (Rosaceae).
*Cydonia oblonga* P. Miller (quince) (Rosaceae).
*Sirex noctilio*: *Pinus patula* Schlechtendal & Chamisso (patula pine) (Pinaceae).
*Pinus radiata* David Don (radiata pine) (Pinaceae).

For endemic species:
*Athalia incomta*: *Selago dinteri* Rolfe (Scrophulariaceae, Lamiales); Opitz *et al.* (2012).
*Athalia marginipennis*: Brassicaceae; Opitz *et al.* (2012).
*Athalia ustipennis*: Brassicaceae; Opitz *et al*. (2012).
*Arge capensis*: *Geranium* sp. (Geraniaceae); Pasteels (1953).
*Arge cochraneae*: *Geranium* sp. (Geraniaceae); Pasteels (1953).
*Arge dirce*: *Diospyros lycioides* Desfontaines (bluebush, red star-apple) (Ebenaceae).
*Arge taeniata*: *Pelargonium* sp. (Geraniaceae); Prinsloo (1985).

# 8. Morphology of Symphyta

## 8.1 Morphology of adults

The suborder Symphyta (sawflies and woodwasps) is a paraphyletic assemblage comprising the structurally more “primitive” Hymenoptera (Smith 1995). The adults of Symphyta share a number of plesiomorphic character states. There is no “wasp-like” constriction at the base of the abdomen as in ants, wasps and bees (suborder Apocrita). The abdomen is broadly attached to the thorax, without this marked flexibly joined constriction between the first and second abdominal segments (Figs 38, 39). In Apocrita the first tergum of abdomen is broadly and immovably fused with metanotum and laterally with metapleuron (propodeum).

The trochanters appear to be two-segmented, because the basal ends of the femora resemble a second segment (trochantellus) of trochanter (Fig. 40D). Especially the fore wings have many veins and numerous enclosed cells (Figs 41A-N), in the smaller hind wings the number of enclosed cells is significantly

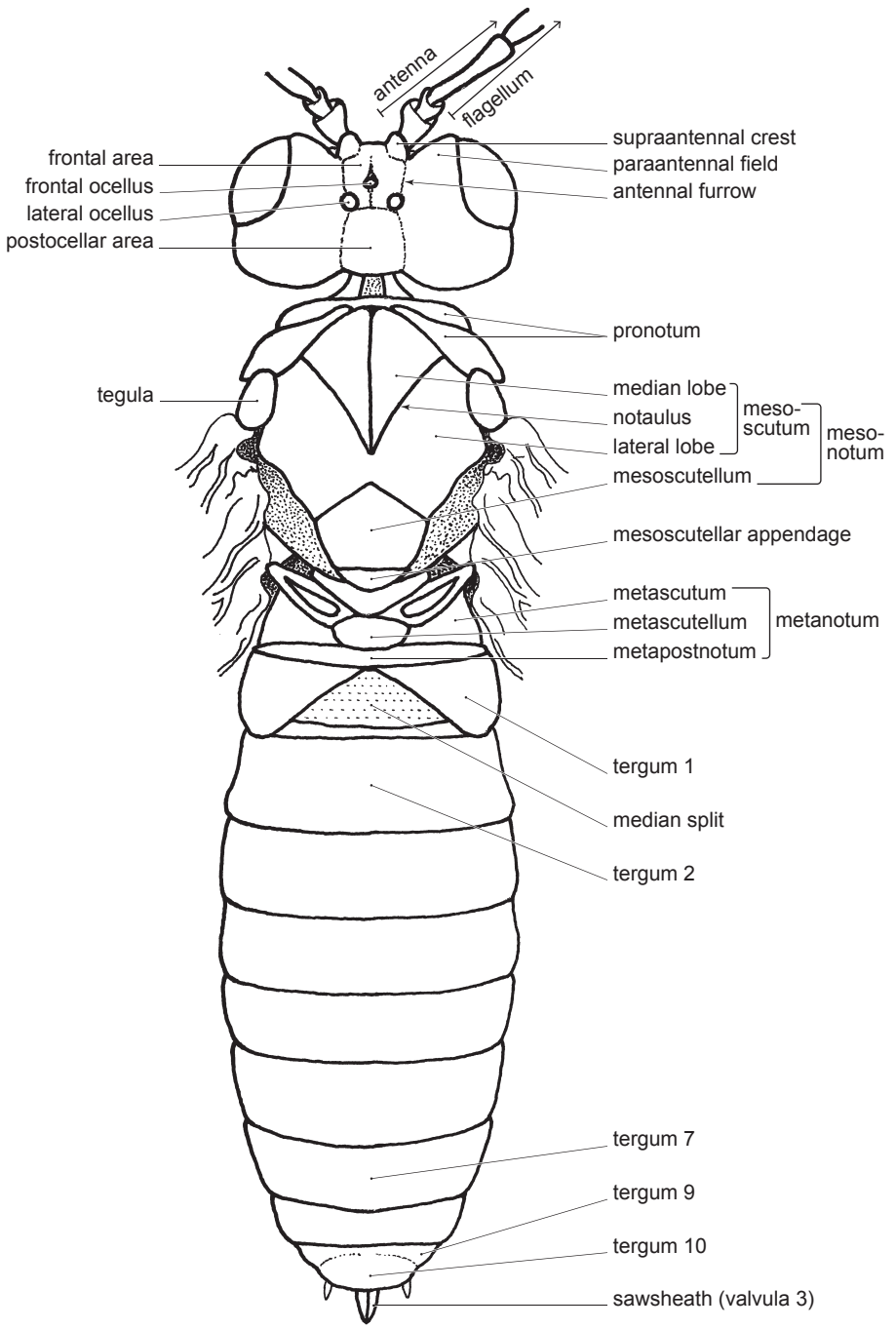


**Fig. 38.** Schematic depiction of the body of a hypothetical Tenthredinidae species (dorsal aspect), modified after Viitasaari (2002).

reduced. In addition to this, cenchri (Fig. 38) are presented on the metascutum, with the exception of the superfamily Cephoidea (family Cephidae), currently comprising only two species, occurring in Madagascar (Muche 1981).

The females of the superfamily Tenthredinoidea possess a saw-like ovipositor (lancet) which facilitates the insertion of eggs into plant tissue, hence the common name sawflies. Furthermore, two apical spines of the fore tibia are present. From the Tenthredionidea in the Afrotropical Region only the families Argidae and

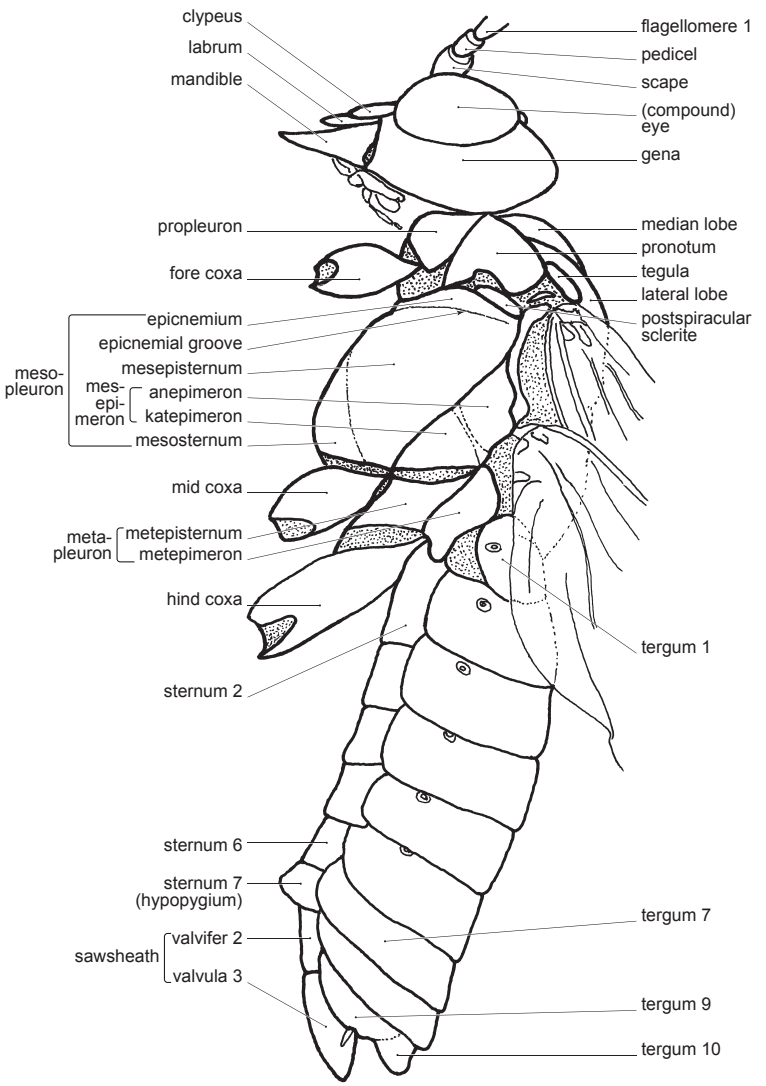


**Fig. 39.** Schematic depiction of the body of a hypothetical Tenthredinidae species (lateral aspect), modified after Viitasaari (2002).

Tenthedinidae are known. The species of all Afrotropical genera are characterized by the absence of an occipital carina.

The worldwide distributed superfamily Siricoidea contains three families – Xiphydriidae, Siricidae and Anaxyelidae. Only the family Siricidae (common names woodwasps and horn-tails) is known in the Afrotropical Region represented by the endemic genus *Afrotremex* and *Sirex noctilio*, which has been introduced into

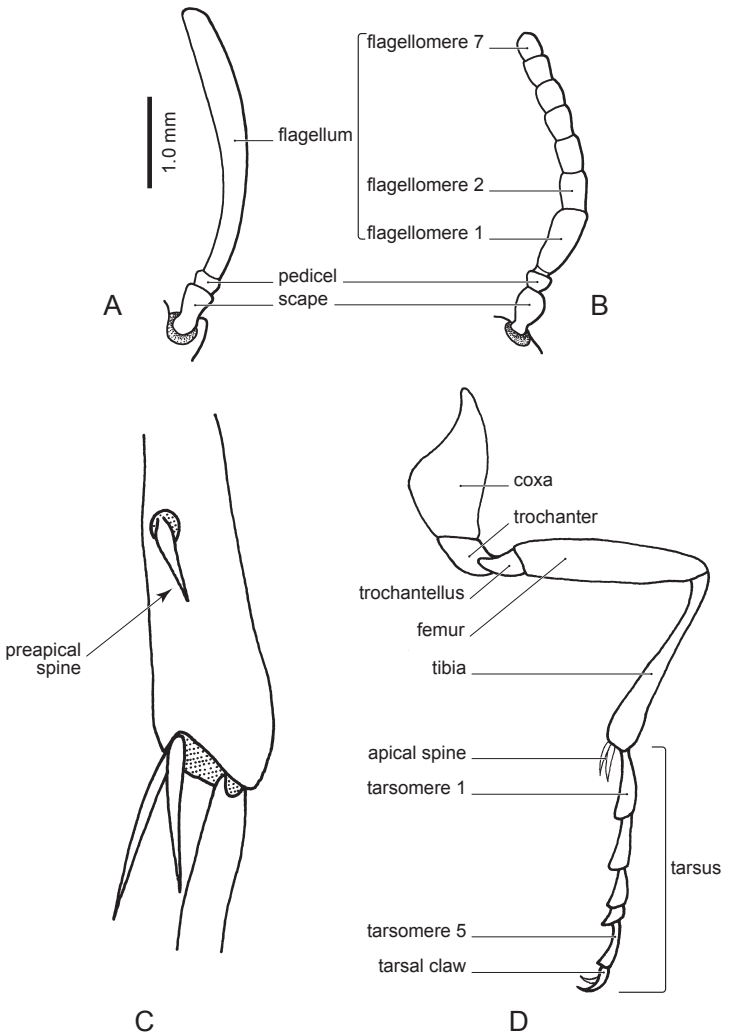


**Fig. 40. A-D**. Schematized antenna: **A**. Argidae (*Arge* sp.). **B**. Tenthredinidae (*Distega* sp.). **C**. Apical half of hind tibia with preapical spine (arrowed) of *Arge* sp. **D**. Hind leg of a hypothetical Tenthredinidae species.

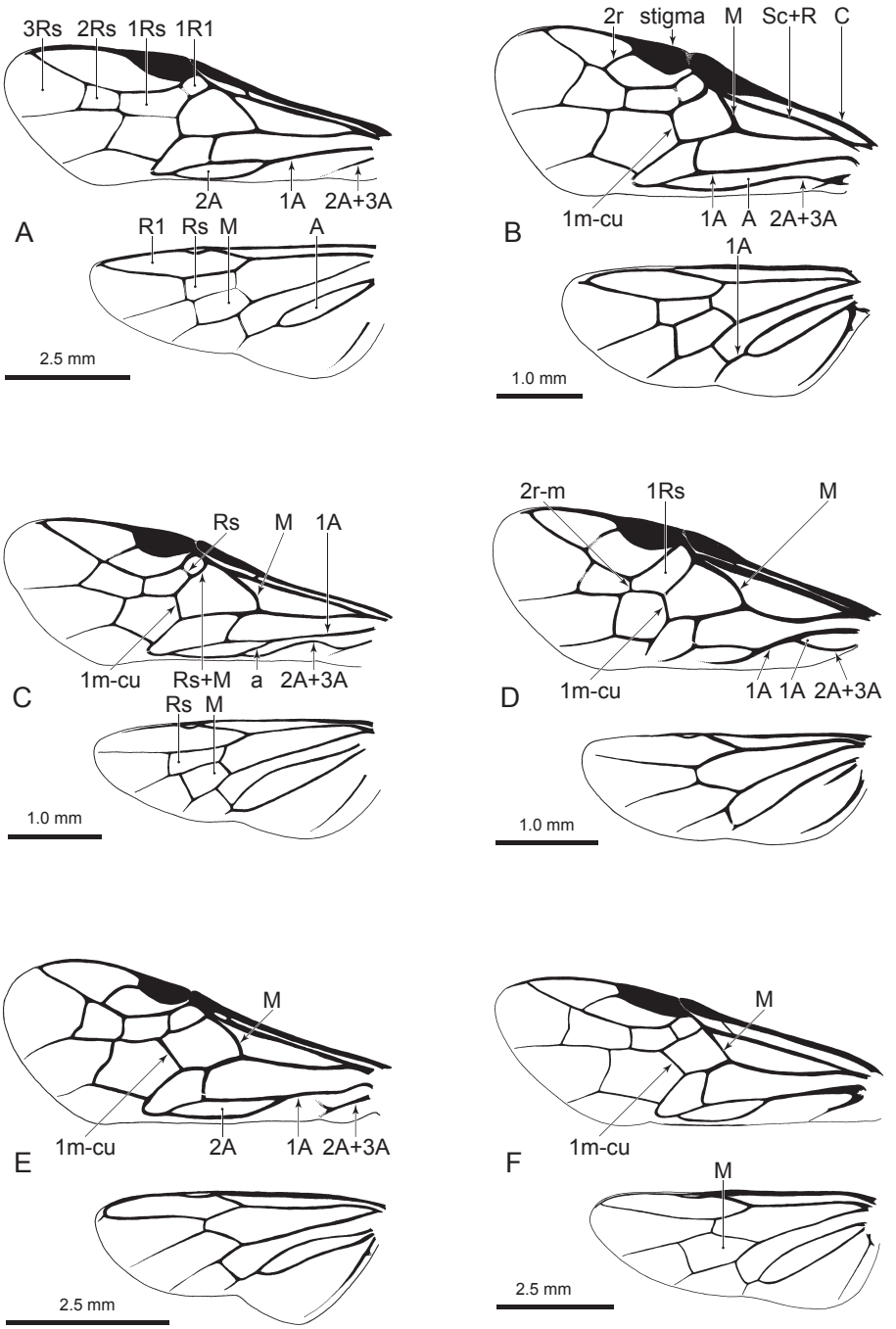


**Fig. 41. A-F**. Fore and hind wing of: **A**. *Nematus oligospilus*. **B**. *Dulophanes obscurus*. **C**. *Caliroa cerasi*. **D**. *Fenusa dohrnii*. **E**. *Durbadnus taegeri*. **F**. *Trisodontophyes diversa*.

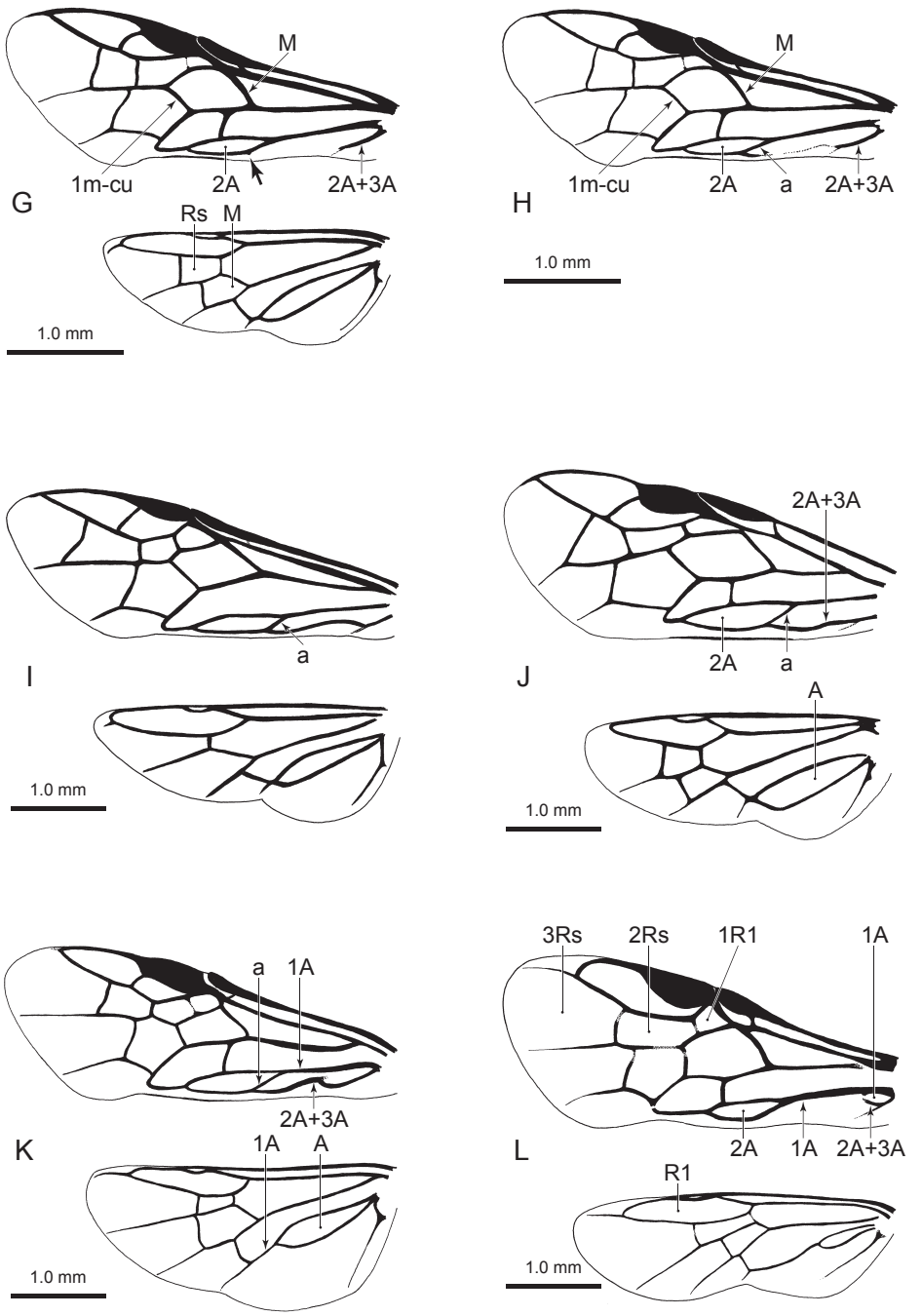


**Fig. 41 (continued). G-L**. Fore and hind wing of: **G**. *Distega* sp. **H**. *Distega bevisi* (fore wing). **I**. *Neacidiophora* sp. **J**. *Xenapates damaraensis*. **K**. *Athalia* sp. **L**. *Triarge namaquaensis*.

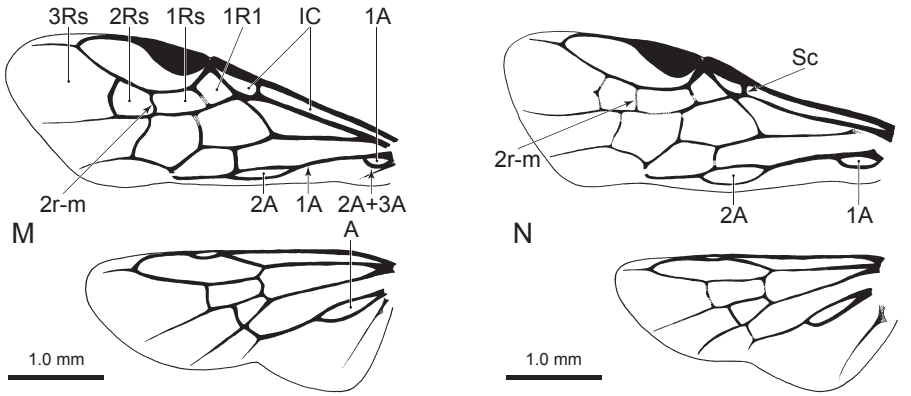


**Fig. 41 (continued). M-N**. Fore and hind wing of: **M**. *Arge* sp.
**N**. *Pampsilota brandbergensis*.

South Africa. The 5 native species of *Afrotremex* are restricted to the west and central African forest areas (Goulet 2014).

In the larger species of Siricidae, which are about 30.0-40.0 mm in length, the ovipositor is long and slender and is used for oviposition in woody tissue, and only one apical spine of the fore tibia is developed.

The fourth Afrotropical sawfly superfamily is Orussoidea with 19 valid species in the region (Taeger *et al*. 2010). The species of the only Afrotropical family Orussidae differ from other Symphyta in their cylindrical cross section, several thorn-like structures on the head around the median ocellus (Figs 161A-C, 162A, B) and in the black to metallic bluish-green colouration. The ovipositor is long and thin, similar to that of Ichneumonidae (Hymenoptera).

**The importance of the genitalia for the identification of species**

Many of the keys of Afrotropical sawflies are based almost entirely on colour. Nevertheless, in all species treated here, except for the introduced species, a thorough examination of the genitalia is essential for an exact species identification. However, the genital morphological features have varying taxonomic value within each genus (Table 1).

**Genitalia of males**

In the genus *Athalia* the penis valves exhibit only a few interspecific differences within the species-groups and cannot always be used for reliable identification. Additionally, the penis valve is often poorly sclerotized, especially the dorsal part. Its shape may be distorted in lateral view as a result of the laterally projecting medio-subapical appendage (Fig. 125I). Conversely, both the shape of the parapenis and

**Table 1:** The morphological structures of the genitalia and their significance for the identification of Afrotropical species, separated according to their genera (XXX - very high; XX - medium to moderately; X - low; O - without).

| | Males | | | Females | | |
|---|---|---|---|---|---|---|
| | Penis valve | Cuspis/Digitus | Harpe/Parapenis | Lancet/Serrulae | Sawsheath | Hypopygium |
| **Argidae** | | | | | | |
| *Arge* | xx(x) | o | x | xxx | xx | o |
| *Pampsilota* | xxx | o | x | xxx | xx | o |
| *Triarge* | x(x) | o | x | xxx | xxx | o |
| **Selandriinae** | | | | | | |
| *Dulophanes* | xxx | o | x | xx | x | o |
| **Blennocampinae** | | | | | | |
| *Distega* | xxx | o | xx | xxx | x | o |
| *Durbadnus* | xxx | o | xx | xxx | x | o |
| *Trisodontophyes* | xxx | o | xx | xxx | x | o |
| **Athaliinae** | | | | | | |
| *Athalia* | x(x) | xxx | xxx | xx(x) | xx | xxx |
| **Allantinae** | | | | | | |
| *Xenapates* | xxx | o | o | xx(x) | x | o |

harpe in ventral view as well as the digitus and cuspis in dorsal or lateral view are characteristic for the species (Fig. 42C) (Abe 1988, Koch 2003, 2006c, 2007).

It is often difficult to make an unambiguous identification based on the lateral view of penis valve for species in the family Argidae. This is because the penis valves are very voluminous in most species and have horn-shaped appendages especially in the middle area. When embedding a microscopic preparation positioning is critical to prevent distortion of the actual shape.

In addition, the inner, more or less large horn projection is surrounded by plenty of muscle tissue and its removal during preparation is often very difficult. The

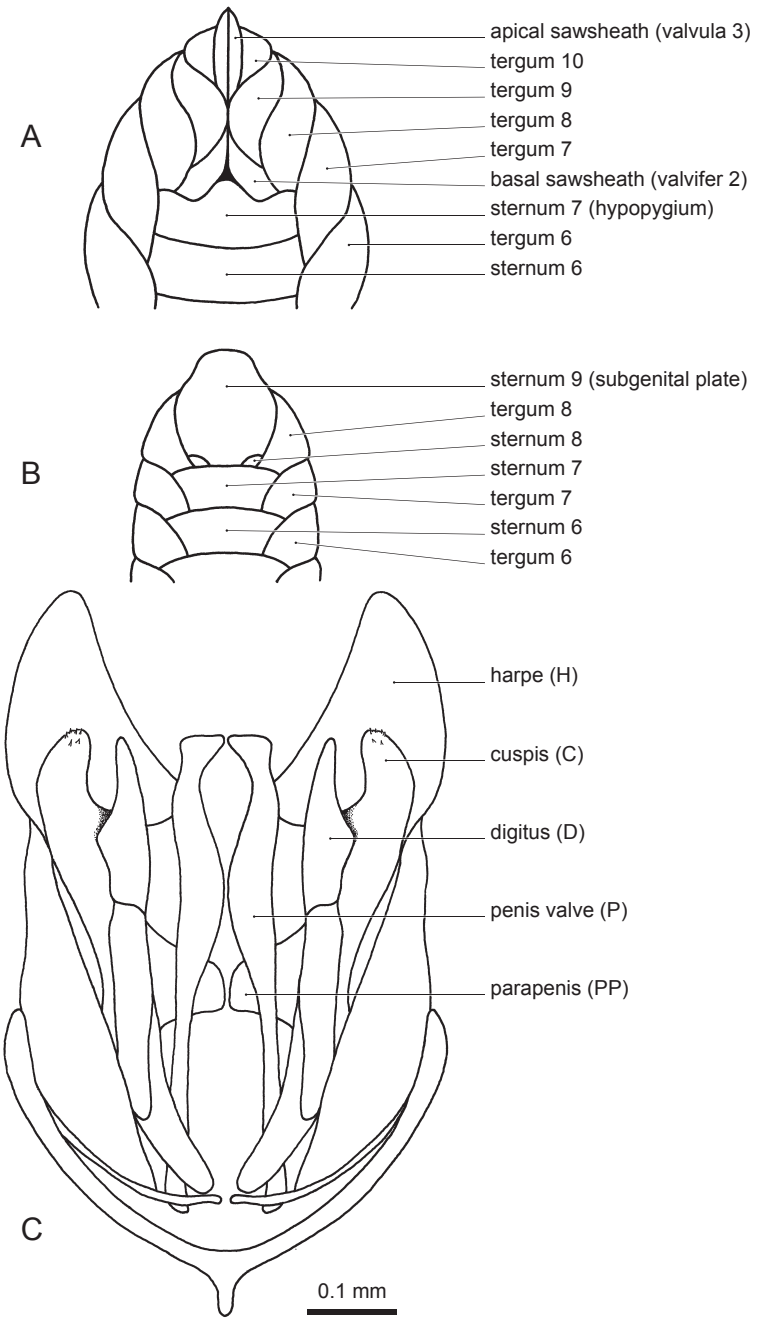


**Fig. 42. A-C**. Schematic depiction of the apical portion of the abdomen of a hypothetical Tenthredinidae species (ventral aspect): **A**. Female. **B**. Male. **C**. Schematic depiction of the capsule of a male of a hypothetical *Athalia* species (dorsal aspect).

extension can break relatively easily and this creates an unrealistic representation. Finally, intraspecific variability, can confound interpretation, particularly when only a small number of specimens are available for examination. This variability is more or less present in all species.

**Genitalia of females**

As presented in Table 1 assessment of the lancet and the shape of the serrulae is critically important in all genera for the correct identification of species.

However, there are some species within the genus *Athalia* that are difficult to distinguish based only on the shape of the serrulae. For example these include the species in the *A. vollenhoveni* species group (Koch 2006c) and the *A. himantopus* species group (Koch 2007). In addition, depending on the age of the females and the frequency of oviposition abrasions of the serrulae are apparent resulting in wear or disappearance of crucial morphological features resulting in an unrealistic picture. However, experience has shown that this phenomenon is relatively rare.

This deficit is primarily compensated by the shape of the posterior margins of the hypopygia (Fig. 42A), which have a very high taxonomic value. Their shape should always be studied in the *Athalia* species. For this purpose the hypopygium must be completely separated and spread over its entire surface, only then will the shape of the taxonomically relevant posterior margin be undistorted and visible for interpretation.

## 8.2 Morphological terms

**Head**

| | | |
|---|---|---|
| Antennal furrow | : | Paired; more or less conspicuously developed lateral furrows of frontal area (Fig. 38). |
| Clypeus | : | Covers more or less the base of mouthparts (Fig. 43A). |
| Epistomal suture | : | Consists of lateral furrows of clypeus and supraclypeal furrow (Fig. 43A). |
| Eye (compound eye) | : | Occupy the greater part of the lateral parts of the head (Fig. 39). |
| Frons | : | Central face between eyes, downwards from the front ocellus including the toruli. |
| Frontal ocellus | : | Anterior ocellus (Fig. 38). |
| Frontal area | : | Between postocellar area and supraclypeal area, includes the ocelli, laterally limited by more or less conspicuously antennal furrows (Fig. 38). |
| Gena | : | Area behind eyes (Fig. 39). |
| Interantennal area | : | Area between toruli (Fig. 43A). |
| Interantennal carina/e | : | Vertical ridges between antennae (Figs 44C, 107B). |

| | | |
|---|---|---|
| Interantennal groove | : | Between supraclypeal and frontal area; very different developed. |
| Intercarinal area | : | Area between interantennal carinae (Figs 44C, 107B). |
| Labrum | : | Is attached under the clypeus (Fig. 43A). |
| Lateral ocellus (ocelli) | : | Two posterior ocelli (Fig. 38). |
| Malar space | : | Area between base of mandible and ventral margin of eye (Fig. 43A). |
| Mandible | : | Paired; belongs to the mouthparts, with apical tooth and sometimes with symmetrically or asymmetrically subapical teeth (Figs 39, 43A); used for biting. |
| Maxillary palp | : | Paired; appendage of the mouthparts (Fig. 134A). |

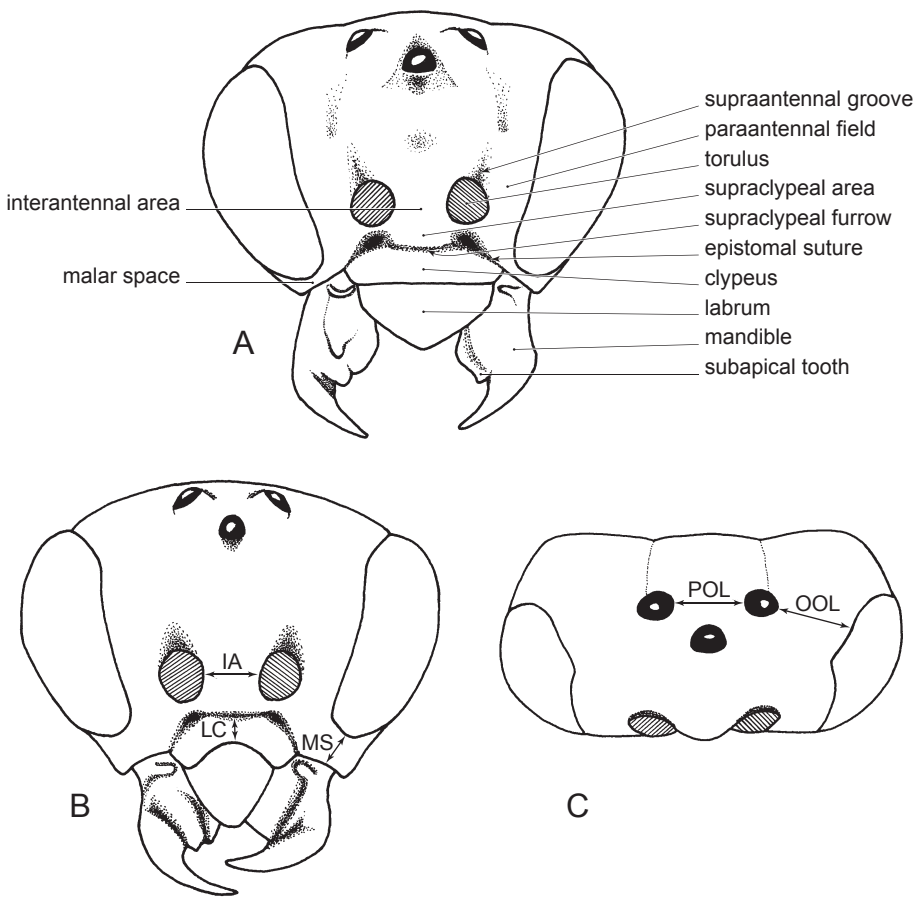


**Fig. 43. A-C**. Schematic depiction of the Head (frontal aspect): **A**. *Athalia himantopus* species group. — **B-C**. *Athalia vollenhoveni* species group, illustrating measurements: **B**. Frontal aspect. **C**. Dorsal aspect.

| | | |
|---|---|---|
| Occipital carina | : | Posterior vertical ridge of gena (see morphology of adults). |
| Paraantennal field | : | Area between eyes and antennae (Figs 38, 43A). |
| Postocellar area | : | Is located on the top of the head and more or less conspicuously separated by lateral furrows and anteriorly by a line between the lateral ocelli (Figs 38). |
| Subapical teeth | : | In some genera symmetrically or asymmetrically presented on the mandibles (Fig. 43A). |
| Supraantennal crest (supraantennal bulge) | : | Paired, a prominent bulge dorso-medial of torulus (Fig. 38). |
| Supraantennal groove | : | Paired; vertical furrow dorsally of torulus, sometimes with antennal furrow connected (Fig. 43A). |
| Supraclypeal area | : | Upwards from supraclypeal furrow to toruli (Fig. 43A). |
| Supraclypeal furrow | : | Dorsal portion of epistomal suture and dorsal margin of clypeus (Fig. 43A). |
| Torulus (toruli) | : | Antennal socket; the scape of the antenna is articulated here (Fig. 43A). |
| Vertex | : | Top of the head, including the ocelli. |

**Antennae**

| | | |
|---|---|---|
| Antenna | : | Consists of scape, pedicel and flagellum (Figs 40A, B). |
| Flagellomere(s) | : | Segments of flagellum (Fig. 40B). |
| Flagellum | : | Very long and unsegmented (Fig. 40A), or segmented in more or less short flagellomeres (Fig. 40B). |
| Pedicel | : | Second segment of antenna (Figs 39, 40A, B). |
| Scape | : | First segment of antenna (Figs 39, 40A, B). |

**Thorax**

| | | |
|---|---|---|
| Anepimeron | : | Dorsal part of mesepimeron (Fig. 39). |
| Epicnemial groove | : | Vertical groove between epicnemium and mesepisternum (Fig. 39). |
| Epicnemium | : | Anterior region of mesepisternum, separated by a more or less conspicuously vertical epicnemial groove (Fig. 39). |
| Katepimeron | : | Ventral part of mesepimeron (Fig. 39). |
| Lateral lobe | : | Part of mesoscutum (Figs 38, 39). |
| Median lobe | : | Part of mesoscutum (Figs 38, 39). |
| Mesepimeron | : | Anepimeron and katepimeron together, posterior part of mesopleuron (Fig. 39). |
| Mesepisternum | : | Anterior part of mesopleuron (Fig. 39). |
| Mesonotum | : | Mesoscutum and mesoscutellum together (Fig. 38). |

| | | |
|---|---|---|
| Mesopleuron | : | Mesepisternum and mesepimeron together (Fig. 39). |
| Mesoscutellar appendage | : | Adjacent to posterior margin of mesoscutellum (Fig. 38). |
| Mesoscutellum | : | Distal part of mesonotum (Fig. 38). |
| Mesoscutum | : | Median and lateral lobes together (Fig. 38). |
| Mesosternum | : | Ventral part of mesepisternum, separated by a more or less conspicuously horizontal suture (Fig. 39). |
| Metanotum | : | Metascutum, metascutellum and metapostnotum together (Fig. 38). |
| Metapleuron | : | Metepimeron and metepisternum together (Fig. 39). |
| Metapostnotum | : | Posterior part of metanotum (Fig. 38). |
| Metascutellum | : | Middle part of metanotum (Fig. 38). |
| Metascutum | : | Anterior part of metanotum (Fig. 38). |
| Metepimeron | : | Dorsal part of metapleuron (Fig. 39). |
| Metepisternum | : | Ventral part metapleuron (Fig. 39). |
| Notaulus (notauli) | : | Furrow between median and lateral lobe of mesoscutum (Fig. 38). |
| Postspiracular sclerite | : | Small sclerite located between pronotum and mesepisternum (Fig. 39). |
| Pronotum | : | Dorsal part of prothorax (Figs 38, 39). |
| Propleuron | : | Lateral part of prothorax (Fig. 39). |
| Tegula (tegulae) | : | Small plates laterally of the thorax covering the junction of the fore wings (Figs 38, 39). |

**Legs**

| | | |
|---|---|---|
| Apical spine (apical spur) | : | In Tenthredinoidea paired spines (spurs) at the apex of tibiae (Fig. 40D). |
| Basal lobe | : | A different shaped enlargement at base of tarsal claw (Figs 137D, 142E, 150D, 155E). |
| Basitarsomere | : | Tarsomere 1, nearest to the tibia (Fig. 40D). |
| Coxa (coxae) | : | (Figs 39, 40D). |
| Femur (femora) | : | (Fig. 40D). |
| Preapical spine (preapical spur) | : | A single spine in the apical half of hind tibia (Fig. 40C). |
| Subapical tooth (teeth) | : | One or two teeth additionally to the apical tooth of tarsal claw (Figs 137D, 150D, 155E). |
| Tarsal claw | : | Paired; simple, or with basal lobe and/or subapical tooth (teeth) (Figs 40D, 44E, 137D, 155E). |
| Tarsomere | : | Segment of tarsus (Fig. 40D). |
| Tarsus (tarsi) | : | Segmented in 5 tarsomeres (Fig. 40D). |

| | | |
|---|---|---|
| Tibia (tibiae) | : | (Fig. 40D). |
| Trochantellus | : | Basal end of the femur; therefore the trochanter seems to be subdivided in two (Fig. 40D). |
| Trochanter | : | The segment between coxa and trochantellus (femur) (Fig. 40D). |

**Wings including morphological abbreviations**

**Wing cells**

**Fore wing** (Figs 41A-N)

| | | |
|---|---|---|
| Anal cell | : | A |
| Basal anal cell | : | 1A |
| First radial cell | : | 1R1 |
| First radial sector | : | 1Rs |
| Intercostal area | : | IC |
| Second anal cell | : | 2A |
| Second radial sector | : | 2Rs |
| Third radial sector | : | 3Rs |

**Hind wing** (Figs 41A-N)

| | | |
|---|---|---|
| Anal cell | : | A |
| Medial cell | : | M |
| Radial cell | : | R1 |
| Radial sector | : | Rs |

**Wing veins**

**Fore wing** (Figs 41A-N)

| | | |
|---|---|---|
| Anal crossvein | : | a |
| Costa (costal vein) | : | C |
| Costal cross vein | : | Sc |
| First anal vein | : | 1A |
| Media | : | M |
| Mediocubital crossvein | : | 1m-cu |
| Radial crossvein | : | 2r |
| Radial sector | : | Rs |
| Radial sector and media | : | Rs+M |
| Radius | : | R |
| Second and third anal vein | : | 2A+3A |

| | | |
|---|---|---|
| Second radiomedial crossvein | : | 2r-m |
| Stigma | : | Strongly sclerotized and broadened apex of costa; palely or darkly coloured. |
| Subcosta and radius | : | Sc+R |

**Hind wing** (Figs 41A-N)

| | | |
|---|---|---|
| First anal vein | : | Petiole of anal cell (1A). |

**Abdomen**

| | | |
|---|---|---|
| Hypopygium | : | Sternum 7 of females (Fig. 42A). |
| Sternum (sterna) | : | Ventral segments 2-7 (females), 2-9 (males) (Figs 39, 42A, B). |
| Subgenital plate | : | Sternum 9 of males (Fig. 42B). |
| Tergum (terga) | : | Dorsal segments 1-10 (Fig. 38). |

**Genitalia of male including morphological abbreviations**

| | | |
|---|---|---|
| Cuspis (C) | : | Part of male genitalia (Fig. 42C). |
| Digitus (D) | : | Part of male genitalia (Fig. 42C). |
| Harpe (H) | : | Part of male genitalia (Fig. 42C). |
| Medio-subapical appendage (MSA) | : | Lateral projection of penis valve (Fig. 125I). |
| Parapenis (PP) | : | Part of male genitalia (Fig. 42C). |
| Penis valve (P) | : | Part of male genitalia (Fig. 42C). |

**Genitalia of female**

| | | |
|---|---|---|
| Lancet | : | Valvula 1 (ventral part of the saw-like ovipositor). |
| Lance | : | Valvula 2 (dorsal part of the saw-like ovipositor). |
| Ovipositor | : | Consists of valvula 1 (ventral) and valvula 2 (dorsal); laterally flanking by the sawsheath. |
| Sawsheath | : | Valvula 3 (apical) and valvifer 2 (basal) are connected; envelope the ovipositor (Figs 38, 39, 42A). |
| Serrula (serrulae) | : | Tooth (teeth) of lancet. |

## 8.3 Sexual dimorphism

The primary sexual morphological differences between males and females are well developed and readily visible on the ventral side of abdomen (Fig. 42A, B). In the females the sawsheath is clearly visible for its entire length as well as the hypopygium, the shape of which is characteristic for many species, particularly those in the genus *Athalia* (Figs 125D, 128D, 130F, 132D, 134E, 136C). The shape of the male subgenital plate situated at the top of the abdomen is sometimes also diagnostic at species level. Except for the apical part of the harpes the actual copulatory organ, comprising the sclerotized genital capsule, is scarcely visible.

The secondary sexual morphological characters such as the shape of the antennae, colouration of the body and pubescence are of variable diagnostic value across the various genera. For example, the flagellum of the male of *Arge* species is much longer and more slender than in the females. Furthermore, the pubescence on the mesepisternum and the mesosternum is sometimes gender-specifically developed, especially for certain *Athalia* species.

With regard to the colouration in certain *Arge* species the proportion of black in the males may be more extended than in the females. Particularly striking is the colour difference in *Sirex noctilio* (Figs 168A, B) the introduced European woodwasp into the study area, in which the female is almost completely black and the abdomen of the male is mostly yellowish-orange. Finally, the females of almost all species are larger than the males.

## 8.4 Morphology of larvae

Larvae are known for only very few Afrotropical sawfly species (Benson 1962, Prinsloo 1985, Visser 2009). Therefore, their morphology is described only briefly in this book. The morphology of sawfly larvae is described in detail by Lorenz & Kraus (1957) and Viitasaari (2002).

Sawfly larvae are superficially similar in appearance to caterpillars, which are the larvae of butterflies and moths. The larvae of sawflies have one simple eye (stemmentum) on each side of the head, whereas in caterpillars of Lepidoptera several eyes are present, and appear as black spots at the lower side margins of the head. Furthermore, the caterpillars of butterflies and moths are distinguished by the possession of five or fewer pairs of abdominal prolegs with crochets, whereas the larvae of most exophytic Tenthredinoidea have six to eight pairs of abdominal legs without crochets. The larvae of most species of Tenthredinidae have prolegs on abdominal segments 2 to 8 and 10 (anal prolegs); the abdominal segments 1 and 9 are free (Fig. 37C). In Nematinae species the prolegs are presented on abdominal segments 2 to 7 and 10.

The larvae of the introduced Heterarthrinae species are different from other Tenthredinidae. The larva of *Caliroa cerasi* is clearly distinguished by the slug-like appearance and its dark or transparent slimy coating (hence the common name). Additionally, the larva is distinguished by possession of a fleshy protuberance, which extends anteriorly from each prothoracic leg.

*Fenusa dohrnii* is one of the leaf-mining sawflies. The larvae are characterized by the more dorsoventrally flattened appearance, smaller thoracic legs and conspicuously reduced prolegs on abdominal segments 2 to 8 and 10.

The morphology of the larvae of the Argidae species is similar to that of Tenthredinidae. The larvae are distinguished by the presence of prolegs on abdominal segment 2 to 6, 2 to 7, 2 to 8 and 10 (Lorenz & Kraus 1957). Argidae

larvae usually have a distinct pad or divergent lobe (emposium) adjacent to the tarsal claw; prothoracic tarsal claw sometimes absent, and abdominal segment with no more than three dorsal annulets.

Larval colour is mostly greenish with small differently coloured spots, larger flecks or stripes, rarely entirely pale without any colour patterns. In many cases the head is differently coloured to the rest of the body. The larva of the invasive *Sirex noctilio* is creamy white, the legs of the thorax are inconspicuous and abdominal legs are absent. A distinctive dark spine is present at the rear of the abdomen.

## 9. Systematic presentation

The systematic arrangement follows the classification structure proposed by Viitasaari (2002) and Taeger *et al*. (2010).

### 9.1 Key to families of Symphyta

1 Antennae inserted on ventral side of head, below ventral margin of eyes and below the posterior margin of clypeus; head with a crown of several thorn-like structures (coronal teeth) around median ocellus (Figs 161A-C, 162A, B); not reported in the study area ……**Orussidae**

1* Antenna inserted in frontal aspect on head, above clypeus and between eyes; head without thorn-like structures around median ocellus (Figs 138A, C) ……2

2 Antenna 3-segmented (3 antennomeres) with only one, very long unsegmented flagellum (Fig. 40A) ……**Argidae**

2* Flagellum of antenna segmented with more than 6 flagellomeres (Figs 40B, 123D, 130B, C, 134C, 137E, 142D, 150C, 155D) ……3

3 Antenna with about 18 flagellomeres (Figs 168A, B); apical tergum of female (Fig. 168A) and apical sternum of male (Fig. 168B) with tube-like projection ……**Siricidae**

3* Antenna mostly with 7-10 flagellomeres, apical sternum and tergum without such projections ……**Tenthredinidae**

#### 9.2 Family Argidae

**Key to genera**

1 Fore wing with crossvein 2r-m absent and cells 1Rs and 2Rs fused, thus there only three submarginal cells with the second very large (Fig. 41L); body usually entirely black ……***Triarge* Forsius**

1* Fore wing with crossvein 2r-m present and cells 1Rs and 2Rs present, thus there are four submarginal cells with the second not conspicuously larger (Fig. 41M); body usually bi-coloured ……2

2 Hind tibia with preapical spine (Fig. 40C ……***Arge* Schrank**
2* Hind tibia without preapical spine ……***Pampsilota* Konow**

## Genus *Arge* Schrank, 1802

**Key to species**

1 Pale body parts orange, at least some of the black body parts with blue metallic lustre; sawsheath conspicuously pincer-shaped without conspicuous coarse bristly setae on interior surface; penis valve as in Figs 55E, 84C, 99B *A. capensis* species group ……2
1* Body black with yellow markings or predominantly yellow with black markings, black body parts without blue metallic lustre, when with metallic lustre then sawsheath robust (Figs 88B, 90B) and interior surface with more or less conspicuous coarse bristly setae, penis valve different to above Figs 48E, 52E, 59 D, 94D ……9

2 Abdomen with orange apex ……5
2* Abdomen entirely black with blue metallic lustre or mid terga and mid sterna orange ……3

3 Abdomen entirely black with blue metallic lustre ……4
3* Abdomen black with blue metallic lustre, mid terga and sterna more or less orange ……***A. elandsbayensis* Koch & Goergen**

4 Thorax predominantly orange, mesosternum black ……
……***A. whiteheadi* Koch & Goergen**
4* Thorax black with blue metallic lustre, only pronotum and tegulae orange ……***A. vannoorti* Koch & Liston**

5 Terga 2-4 orange laterally, mesopleuron and mesosternum black……
……***A. cochraneae* Koch & Goergen**
5* Abdomen without orange markings laterally, at least mesopleuron with orange markings or male ……6

6 Male……7
6* Female ……8

7 Mesoscutum black with blue metallic lustre, median lobe of mesoscutum orange ……***A. capensis* (Klug)**
7* Mesoscutum without orange markings ……***A. namaensis* Koch & Goergen**

8 Thorax nearly entirely orange; lancet with more or less uniformly cigar-shaped trichoid sensilla ……***A. rufocyanea* (Enslin)**
8* Thorax sometimes partly more black coloured, especially on mesopleuron and mesosternum; lancet with long filiform trichoid sensilla ……***A. capensis* (Klug)**

9 Thorax entirely black with conspicuous blue metallic lustre; fore wing with fuscous crossband under stigma ……***A. speciosa*** **(Klug)**
9* Thorax nearly entirely black or predominantly yellow, fore wing without conspicuously infuscate crossband ……10

10 Thorax nearly entirely black; abdomen entirely yellow, at most with small blackish markings ……11
10* Thorax partly or predominantly yellow; abdomen differently coloured ……13

11 Wings sharply bicoloured, basal half slightly flavescent-hyaline and apical half fuscous, costa entirely yellow, intercostal area flavescent-hyaline ……***A. bisignata*** **Konow**
11* Wings uniformly flavescent-hyaline or very slightly infuscate throughout, costa and intercostal area dark brown ……12

12 Wings uniformly flavescent-hyaline throughout; hind tibia black ringed apically; hind coxa predominantly yellow, at least ventral surface …… ……***A. annulipes*** **(Klug)**
12* Wings very slightly infuscate throughout; hind tibia entirely yellow; hind coxa entirely black ……***A. spei*** **(Enslin)**

13 Head more or less yellow, at least postocellar area yellowish ……14
13* Head black or with blue metallic lustre, at most mandible and labrum light brown ……17

14 Head black, only postocellar area yellow; sawsheath in dorsal view broadly and obtusely pincer-shaped apically (Fig. 75C) …… ……***A. kungveldensis*** **Koch & Eardley**
14* Head extensively yellow; sawsheath in dorsal view not pincer-shaped apically ……15

15 Head strongly enlarged behind eyes; legs entirely black with slight blue metallic lustre ……***A. sjoestedti*** **Konow**
15* Head slightly enlarged behind eyes; legs predominantly yellow ……16

16 Head entirely yellow; mesoscutellum black, mid femur broadly black ringed apically ……***A. hereroensis*** **Koch & Goergen**
16* Interocellar area and postocellar area black; mesoscutellum yellow; mid femur entirely yellow ……***A. meyi*** **Koch**

17 Legs entirely black ……22
17* Legs partly yellow ……18

18 Only fore tibia and fore basitarsomere yellow; hind tibia distinctively widened apically (Fig. 51A) ……***A. bensoni*** **Pasteels**
18* Legs extensively yellow; hind tibia not distinctively widened apically ……19

19 Costa entirely yellow ……20
19* Costa more or less brownish, apex adjacent to stigma always brown ……21

20 Anal vein 1A of fore wing and veins of hind wing brownish; abdomen yellow with more or less broad black median stripe on dorsal surface; in ♀♀ the median stripe reduced to median spots; penis valve as in Figs 59D, E ……***A. deckerti* Koch**
20* Veins of basal halves of fore and hind wings entirely yellow; abdomen yellow with black patch on apical half on dorsal surface; penis valve as in Fig. 71 ……***A. iota* Pasteels**

21 In ♂♂ at least mid and hind femur entirely black; in ♀♀ hind femur more or less black; apex of sawsheath black; sawsheath in dorsal view pincer-shaped (Fig. 66B); penis valve as in Fig. 66E ……***A. furvipes* Konow**
21* In ♂♂ and ♀♀ all femora entirely yellow; sawsheath yellow; sawsheath in dorsal view not pincer-shaped, moderately rounded apically (Fig. 94A); penis valve as in Fig. 94 D ……***A. stuhlmanni* (Kohl)**

22 Abdomen entirely black with slight blue metallic lustre …… ……***A. krabbefonteinensis* Koch & Goergen**
22* Abdomen more or less yellow ……23

23 Dorsal half of mesopleuron and lateral lobe of mesoscutum yellow…… ……***A. taeniata* (Klug)**
23* Mesopleuron and mesonotum entirely or nearly entirely black or black with blue metallic lustre ……24

24 At least some of the black body parts with blue metallic lustre; sternum 9 of ♂♂ black; sawsheath of ♀♀ entirely black, in dorsal view compact, moderately rounded apically, interior surface slightly convex with coarse, very short bristles (Fig. 63B) ……***A. dirce* (Kirby)**
24* Black body parts without blue metallic lustre, sternum 9 of ♂♂ yellow; sawsheath of ♀♀ yellow with black apex, pincer-shaped (Figs 45B, 77B, 82B) ……25

25 Costa and subcosta nearly entirely yellow, only apex adjacent to stigma brown, intercostal area flavescent-hyaline; serrulae at centre moderately flattened, obtusely angular on anterior edge (Fig. 45D); penis valve as in Fig. 45E ……***A. angulifera* Pasteels**
25* Costa and subcosta predominately brownish with yellow base, intercostal area infuscate; serrulae at centre flattened or prominent, each with rounded anterior edge (Figs 77D, 82 D); penis valve as in Figs 77E, 82E ……26

26 Head slightly enlarged behind eyes; dorsal surface of abdomen medially with narrow black longitudinal stripe; serrulae at centre flattened (Figs 82C, D); penis valve as in Fig. 82E ……***A. montana* Koch & Goergen**

26* Head parallel behind eyes; dorsal surface of abdomen of ♂♂ nearly entirely black; ♀♀ medially with somewhat broader black longitudinal stripe; serrulae at centre prominent (Figs 77C, D); penis valve as in Fig. 77E ……***A. langebergensis* Koch & Goergen**

### Genus *Pampsilota* Konow, 1899

**Key to species**

1 Thorax entirely black; femora black, tibiae and tarsi pale …… ……***P. luederitzensis* Koch**
1* Pronotum and tegula yellow; legs black, fore tibia and tarsus brownish ……***P. brandbergensis* Koch**

### Genus *Triarge* Forsius, 1931

**Key to species**

1 Abdomen entirely black or black with metallic lustre ……2
1* Abdomen black with yellow apex……***T. flavoapicalis* Koch**

2 ♂♂ ……3
2* ♀♀ ……5

3 Supraclypeal area rounded in lateral view (Fig. 107A) …… ……***T. citrusdalensis* Koch**
3* Supraclypeal area more or less flat or slightly rounded ……4

4 Parapenis roundly excised for about a half of its length (Fig. 116G) ……***T. namaquaensis* Koch**
4* Parapenis circularly excised for about a third of its length (Fig. 107G) ……***T. mosselbayensis* Koch**

5 Sawsheath in dorsal view very broadly forcipated (Figs 114D, 115D) ……6
5* Sawsheath in dorsal view more or less narrowly forcipated (Figs 107D, 110D, 112D, 116D, 119D, 121D, 122D) ……7

6 Tibiae more or less yellowish. Namibia ……***T. karooensis* Koch**
6* Tibiae black. South Africa ……***T. mosselbayensis* Koch**

7 Abdomen black without blue metallic lustre ……8
7* Abdomen black with blue metallic lustre ……10

8 Interantennal carina very short (Fig. 122B) ……***T. winterhoekensis* Koch**
8* Interantennal carina longer ……9

9 Sawsheath in dorsal view slightly broadly forcipated (Fig. 116D); serrulae sharp, hook-like (Figs 116E, F) ……***T. namaquaensis* Koch**

9* Sawsheath in dorsal view narrowly forcipated (Fig. 110D); serrulae less acute and less hooked (Figs 110E, F) ……***T. driehoekensis* Koch**

10 Interantennal carina short, extending about ¼ way to clypeus ……11

10* Interantennal carina distinctly longer, extending about half way to clypeus ……***T. plumbea* Forsius**

11 Fore tibia light brown ……***T. citrusdalensis* Koch**

11* Fore tibia blackish ……***T. nigra* Koch**

## 9.3 Family Tenthredinidae

### Key to subfamilies

1 Fore wing with radial crossvein 2r present (Figs 41B-K) ……2

1* Fore wing with radial crossvein 2r absent (Fig. 41A) ……**Nematinae**

2 Fore wing with anal cell present, anal veins (2A+3A) completely outlined, without anal crossvein (a) (Fig. 41B); anterior part of mesepisternum with epicnemium, separated by a more or less conspicuous epicnemial groove (Fig. 39) ……**Selandriinae**

2* Fore wing with anal cell 2A petiolate (1A), 2A+3A reduced to a stub (Figs 41E-H) or curved up to anal vein 1A (Figs 41D, E); or 2A+3A complete and anal crossvein (a) present (Figs 41C, I-K); mesepisternum without epicnemium …3

3 Fore wing with vein M and crossvein 1m-cu more or less convergent (Figs 41C, D) ……**Heterarthrinae**

3* Fore wing with veins M and 1m-cu parallel (Figs 41E-K) ……4

4 Fore wing with anal cell 2A petiolate (1A), 2A+3A reduced to a stub (Figs 41E-H) ……**Blennocampinae**

4* Fore wing with anal cell entirely present, 2A+3A completely outlined, anal crossvein (a) present (Figs 41I-K) ……5

5 Antenna 10-12-segmented, apical flagellomere sometimes indistinctly separated (Fig. 123D); tarsal claws simple (Fig. 123E) ……**Athaliinae**

5* Antenna 9-segmented, distal flagellomeres conspicuously separated (Fif. 40B); tarsal claws with enlarged basal lobe (Fig. 142 E) or with basal lobe and apical tooth (Fig. 137D) ……**Allantinae**

## Subfamily Athaliinae

### Genus *Athalia* Leach, 1817

**Key to species groups of *Athalia* in the Afrotropical Region**

1 Clypeus elongate medially and rounded in front (Fig. 123A) ……
……***Athalia* "*incomta*" species group**
1* Clypeus short medially and more or less truncate or conspicuously excised in front ……2

2 Clypeus short medially and truncate to subtruncate in front (Fig. 123B) ……***Athalia himantopus* species group**
2* Clypeus very short medially and conspicuously excised in front (Fig. 123C) ……***Athalia vollenhoveni* species group**

**Key to *Athalia* species**

1 Clypeus elongate medially and rounded in front ……2
1* Clypeus short medially and more or less truncate to subtruncate in front ……***A. marginipennis* Enderlein**

2 Mesonotum nearly entirely black; maxillary palp not extensively elongated …3
2* Mesonotum yellow with black median lobe of mesoscutum; maxillary palp extensively elongated (Fig. 134A.) ……***A. turneri* Forsius**

3 Fore and mid tibia entirely yellow, only hind tibia black apically; pronotum and tegula yellow or black ……4
3* Mid and hind tibia black apically; pronotum and tegula black ……
……***A. ustipennis* Mocsáry**

4 Male ……5
4* Female ……7

5 Pronotum, tegula and mesopleuron yellow ……***A. brevicornis* Benson**
5* Pronotum, tegula and mesopleuron more or less black ……6

6 Pronotum, tegula, mesopleuron and dorsal margin of mesosternum entirely black ……***A. incomta* Konow**
6* Pronotum and mesopleuron partly blackish, tegula yellow, mesosternum entirely yellow ……***A. maraisi* Koch**

7 Pronotum, tegula and mesopleuron yellow; sawsheath in dorsal view conspicuously enlarged apically (Fig. 130E) ……***A. maraisi* Koch**
7* Pronotum, tegula, mesopleuron and dorsal margin of mesosternum entirely black; sawsheath in dorsal view pointed apically (Fig. 128C) ……***A. incomta* Konow**

## Subfamily Allantinae

**Key to genera**

1 Hind wing with cells Rs and M absent (Fig. 41I) ……***Neacidiophora*** **Enslin**
1* Hind wing with cells Rs and M (Fig. 41J) present ……***Xenapates*** **W.F. Kirby**

### Genus *Neacidiophora* Enslin, 1911

*Neacidiophora brevicornis* Pasteels, 1954

### Genus *Xenapates* W.F. Kirby, 1882

**Key to species**

1 Mesopleuron entirely black ……2
1* Mesopleuron with white marking ……3

2 Head entirely black ……***X. beateae*** **Koch**
2* Labrum, anterior margin of clypeus, base of mandible and a spot at genal orbit white ……***X. damaraensis*** **Koch**

3 Dorsal half and a narrow posterior margin of genal orbit white; terga black with only narrow white posterior margins ……***X. eardleyi*** **Koch**
3* Genal orbit entirely black; mid terga additionally with more or less large dirty whitish medial spot, in male fused to a large pale longitudinal patch (Fig. 147C) ……***X. similis*** **Benson**

## Subfamily Blennocampinae

**Key to genera**

1 Hind wing with cell M (Fig. 41F) or both Rs and M (Fig. 41G) present ……2
1* Hind wing with cells Rs and M absent (Fig. 41E) ……***Durbadnus*** **Pasteels**

2 Hind wing only with cell M present (Fig. 41F); tarsal claw tridentate with large basal lobe (Fig. 155E); mesepisternum without transverse groove or suture …… ……***Trisodontophyes*** **Enslin**
2* Hind wing with Rs and M both present (Fig. 41G); tarsal claw with one inner tooth and enlarged basal lobe (Fig. 150D); upper half of mesespisternum separated from lower by a transverse groove or suture (Fig. 149A) ……***Distega*** **Konow**

### Genus *Distega* Konow, 1904

**Key to species**

1 Thorax entirely black, abdomen and femora yellow ……***D. montium*** **Konow**