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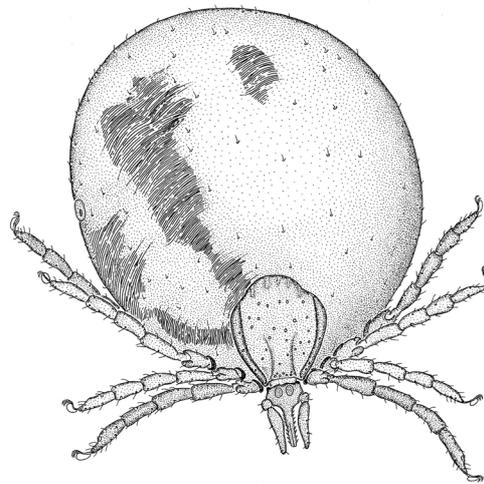
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Ticks of Australia.

The species that infest domestic animals and humans

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STEPHEN C. BARKER & ALAN R. WALKER

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Book cover: *Ixodes holocyclus* (paralysis tick, unfed female)

Title page picture: *Ixodes holocyclus* (paralysis tick, engorging female)

Table of contents

Abstract	4
Introduction and glossary	4
Introduction	4
How to use the guide to identify species, and the glossary	5
Glossary	6
Biology of ticks and methods for identification	19
Evolutionary relationships of ticks to other animals	19
Feeding of ticks in relation to animal and human health	20
Host finding	21
Tick reproduction and life-cycles	21
Mating	21
Life-cycle of three-host ticks	21
Life-cycles of one and two-host ticks	23
Life-cycles of argasid ticks	24
Tick ecology	24
Habitats and hosts	24
How to sample ticks	25
Collection of ticks from hosts	25
Tick sampling and forceps	27
Collection of ticks from vegetation and other environments	27
Preservation and labelling	27
Observing ticks	27
External structure	28
Genera and species of ticks	28
Procedure to identify ticks	28
Genus	28
Species	29
Character matrices	30
Further reading	35
Species accounts	45
<i>Argas persicus</i> (poultry tick)	45
<i>Argas robertsi</i> (Robert's bird tick)	48
<i>Ornithodoros capensis</i> (seabird soft tick)	53
<i>Ornithodoros gurneyi</i> (kangaroo soft tick)	57
<i>Otobius megnini</i> (spinose ear tick)	61
<i>Amblyomma triguttatum</i> (ornate kangaroo tick)	63
<i>Bothriocroton auruginans</i> (wombat tick)	69
<i>Bothriocroton hydrosauri</i> (southern reptile tick)	73
<i>Haemaphysalis bancrofti</i> (wallaby tick)	80
<i>Haemaphysalis longicornis</i> (bush tick)	84
<i>Ixodes cornuatus</i> (southern paralysis tick)	89
<i>Ixodes hirsti</i> (Hirst's marsupial tick)	95
<i>Ixodes holocyclus</i> (paralysis tick)	100
<i>Ixodes tasmani</i> (common marsupial tick)	112
<i>Rhipicephalus (Boophilus) australis</i> (Australian cattle tick)	118
<i>Rhipicephalus sanguineus</i> (brown dog tick)	123
Summation and future prospects	127
Acknowledgements	128
References	128

Abstract

The book *Australian Ticks* by F.H.S. Roberts (1970) is a land-mark in Australian tick biology. But it is time for a new and improved book on the ticks of Australia. The present book has identification guides and accounts of the biology and diseases associated with the 16 species of ticks that may feed on domestic animals and humans in Australia. These comprise five argasid (soft) ticks: *Argas persicus* (poultry tick), *Argas robertsi* (Robert's bird tick), *Ornithodoros capensis* (seabird soft tick), *O. gurneyi* (kangaroo soft tick), *Otobius megnini* (spinose ear tick); and 11 ixodid (hard) ticks, *Amblyomma triguttatum* (ornate kangaroo tick), *Bothriocroton auruginans* (wombat tick), *B. hydrosauri* (southern reptile tick), *Haemaphysalis bancrofti* (wallaby tick), *H. longicornis* (bush tick), *Ixodes cornuatus* (southern paralysis tick), *I. hirsti* (Hirst's marsupial tick), *I. holocyclus* (paralysis tick), *I. tasmani* (common marsupial tick), *Rhipicephalus (Boophilus) australis* (Australian cattle tick) and *R. sanguineus* (brown dog tick). We use an image-matching system to identify ticks, much like the image-matching systems used in field-guides for birds and flowers. Ticks may be identified by drawings that emphasise unique matrices of uniformly defined morphological characters that, together, allow these 16 ticks to be identified by morphology unequivocally. The species accounts have seven sections: (i) General; (ii) Differential diagnosis; (iii) Hosts; (iv) Life-cycle and seasonality; (v) Disease; (vi) Habitat and geographic distribution; (vii) Genes and genomes; and (viii) Other information. There are 71 figures and tables, including a glossary character matrices, drawings of life-cycles, drawings of genera, species, and colour photographs of tick biology.

Key words: Ixodida, Ixodidae, Argasidae, ticks, illustrated diagnostic guide

Introduction and glossary

Introduction

Anyone who has tried to identify an Australian tick has struggled with the dichotomous keys and illustrations in *Australian Ticks* (Roberts 1970). Hoogstraal (1971) lamented that "owing to the lack of a professional illustrator, many figures in the book [Roberts 1970] are inadequate for critical taxonomic differentiation". We have taken the first step to remedy this with drawings of the 16 species of ticks that may feed on humans and our domestic animals.

Hoogstraal (1971) also lamented that the "slight amount of available biological information on Australian ticks is disappointing to many parasitologists, zoologists and biologists". We have endeavoured to remedy the amount of biological information that is available on Australian ticks by critical appraisal of the 500 or so papers that present information and ideas on these 16 species of ticks, from the first paper on the biology of an Australian tick by Bancroft (1884) on paralysis in dogs caused by *Ixodes holocyclus*, to papers published in April 2014; 170 years later. In our species accounts, we have tried to set down what the tick looks like, how it lives and reproduces, where it lives, and the effects the tick might have on its hosts. Nonetheless our species accounts are woefully inadequate in places. For example, there are few published precise site-records and host-records for many of the 16 species of ticks. We hope our work will highlight the gaps in knowledge of the biology of those ticks and thus provide pointers for students and others hunting for research topics.

The common names of ticks and their hosts are usually instructive to us, and often more instructive to non-zoologists than the scientific names of the hosts and of the families of hosts, so we have included the common names of Australian ticks (Carne *et al.* 1987), mammals (Van Dyke & Strahan 2008), birds (Pizzey & Knight 2010) and reptiles (Wilson & Swan 2010). Where we give the number of species in a genus or family, these numbers come from Guglielmone *et al.* (2010) which is the list of valid species, as at 2010, that was agreed to by the experts in our field.

Despite its inadequacies, *Australian Ticks* (Roberts 1970) has served Australian parasitologists, veterinarians, medical workers and wildlife biologists well for half a century. Frederick Hugh Sherston Roberts (1901–1972) stands tall as *the* father of Australian tick biology. "Bob" Roberts suffered a stroke and was gravely ill when *Australian Ticks* was published. Sadly, he was too ill to recognise his book when it was shown to him in about 1970 (J.F.A. Sprent pers. comm. to SB). Roberts also published *Insects Affecting Livestock* (1952) which was for many years a standard textbook in Australia and elsewhere. We salute the father of Australian tick biology.

How to use the guide to identify species, and the glossary

We use a simple two-step process for identifying the ticks of domestic animals and humans in Australia. The first step leads to the name of the genus to which a tick belongs, e.g. *Ixodes*, and its sex. The second step identifies the tick at the species level, e.g. *Ixodes holocyclus*. These steps involve simple visual matching of specimens to the photographs and drawings, whilst examining the tick with a dissecting microscope or hand lens.

The first part of this guide explains how to use the guide and includes a glossary of the morphological and technical terms used to define the different genera and species of ticks. The morphological terms are the characters of body shape, size and texture used for identification; each character has two or more contrasting character-states. The same words for the terms are used to label the drawings that illustrate each genus and species. All character-states in the guide are illustrated in the glossary, except for colours of ticks. The technical terms relate to taxonomy, climate, etc. The glossary is placed first because it is essential for the identification process and it is easiest to find here. We outline general information on tick biology that is useful in identifying ticks, including tick life-cycles, and their veterinary and medical importance. We also explain how to collect and preserve ticks and the procedures used to examine ticks in the laboratory. *This section should be read completely by anyone without a detailed knowledge of ticks.* Then we take the user through the two steps needed to identify tick species.

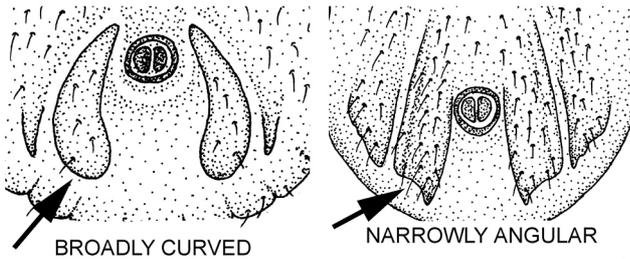
A holistic system is used where all information is made available simultaneously. This is not the usual type of identification key (= dichotomous key). Rather, groups of strictly defined characters are used and each character exists in two or more states. Take for example: the morphological feature 'cervical groove'. There are two diagnostic characters for this feature: depth and length. The character 'cervical grooves length' has two character-states, 'short' and 'long'. These are defined in the glossary with an illustration relating 'short' and 'long' to the size of the scutum of the tick. The two states for depth are indicated by shading on the illustration. Each species has a unique combination of character-states. For each species, this combination of character-states is listed below the set of drawings for the species. For very similar species there will be only one or two differences in the combinations of character-states. In summary, it is a *combination of character-states* that needs to be checked against the specimen when identifying a tick to genus or species. All character-states are defined in the annotated illustrated glossary. Although the guide does not use a dichotomous key, the character-states are the same types of features used in dichotomous keys by other authors. The advantage of the format of this guide is that specimens can be compared directly with drawings showing the overall combination of all character-states together, as seen on the tick. For identification, it is important to use all the information provided including the complete set of character-states for each species, and the clinical context such as geographic distribution and host. It is important to examine whenever possible both female and male specimens, and a large sample of these if possible.

This guide is selective; it covers only 16 species recognized by us as important to the health of domestic animals and humans in Australia. Ticks of livestock, horses, poultry, dogs and cats are covered but not all ticks of wildlife and zoo animals. Ticks of importance to human health in Australia are, by natural coincidence, within this selection. Adult ticks only are covered (with the exception of *Otobius megnini*) because immature ticks are difficult to identify to species. Some character-states can be difficult to distinguish and may be highly variable. Base your identifications on as many character-states as possible. If you remain unsure of an identification consider that your specimen could be a species that feeds mainly on wild animals and is not covered by this guide. Consult the dichotomous key of Roberts (1970) or find an expert to help.

GLOSSARY

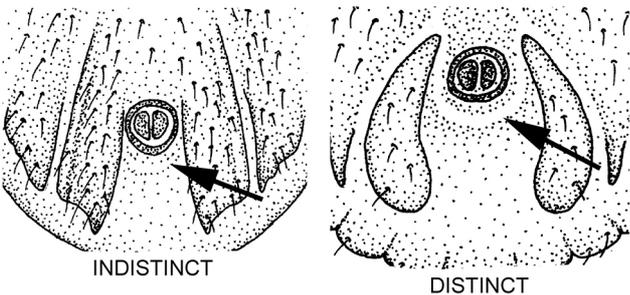
Acari: the taxonomic group that contains the ticks and mites. Ticks are very similar in structure and biology to mites but ticks are larger and exclusively blood feeding parasites. There are many different groups of mites but only one group of ticks, known as the Ixodida.

Adanal plate shape (also see below, Anal plates): variations in a pair of hard plates (or sclerites) on the ventral surface of some male ticks, closest to either side of the anus. They may be **broadly curved at the posterior end**, or **narrowly angular at the posterior end**.

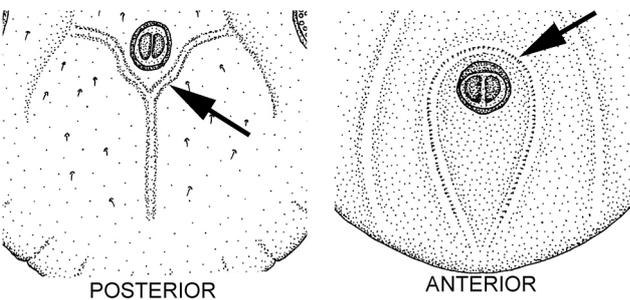


Alloscutum (compare with Scutum): the posterior, non-hardened, part of the body wall of female ixodid ticks.

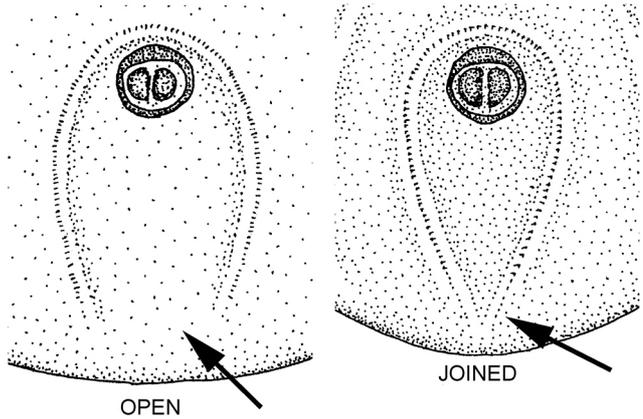
Anal groove: this is a groove in the body wall partially surrounding the anus. It may be **indistinct**, or **distinct**.



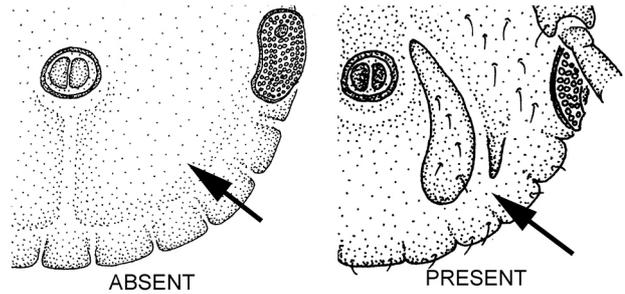
Anal groove position: where the anal groove occurs it forms a line **posterior to the anus**, but in the genus *Ixodes* it forms a line **anterior to the anus**.



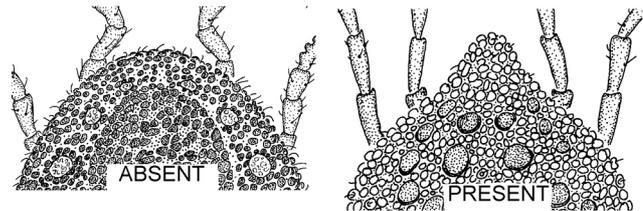
Anal groove shape: the anal groove in *Ixodes* females may be either **open at posterior**, or **joined at posterior** to form a point.



Anal plates (sclerotized or ventral plates): males of some genera have hardened (sclerotized) areas of integument aligned in pairs around the anus. These plates are either entirely **absent** or some or all of them are **present**. They comprise the adanal, accessory and sub-anal plates. Also some *Amblyomma* males have small plaques of integument aligned with the ventral posterior margin. *Ixodes* males have broad plates of sclerotized integument covering most of their ventral surface, of various names.



Anterior projection of body: in *Ornithodoros* ticks this is **absent**, or **present**.

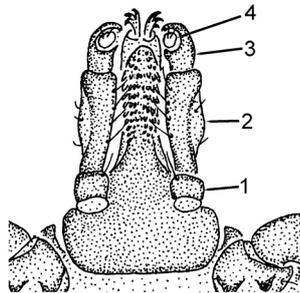


Argasidae: within the Acari the ticks are in the order Ixodida, and within this there are two main families, the Argasidae and the Ixodidae. Argasid ticks are also known as the soft ticks because they lack the hardened scutum of the ixodid ticks. However, soft ticks are tough and well protected against environmental stress.

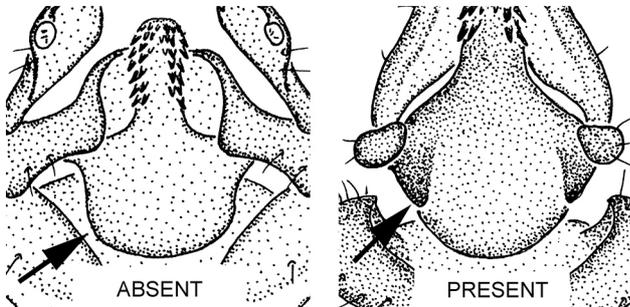
Arthropoda: the taxonomic group (a phylum) that contains ticks, mites, spiders, crustaceans and similar animals. They have an exoskeleton which is divided into segments and limbs with moveable joints moved by internal muscles. Segmentation in the main body of ticks is greatly reduced compared to insects.

FIGURE 1a. Glossary: Acari, to Arthropoda.

Article (of palps): the palps are parts of the mouthparts, like miniature limbs, mainly for sensing the best site to feed. Each palp consist of four segments, known as articles, counted from the base outwards. Sometimes only two of these are easily visible or articles 2 and 3 are fused.

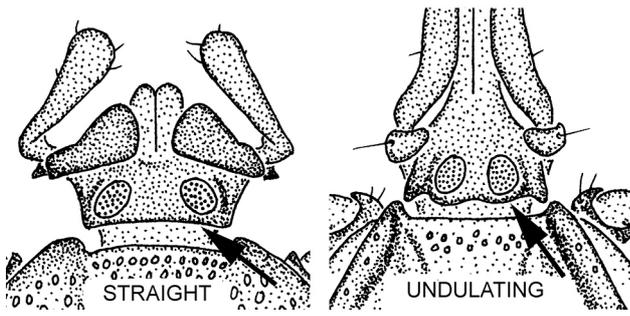


Auriculae: in *Ixodes* the ventral surface of the basis may have lateral bulges or spurs called auriculae. These may be **absent**, or **present**.

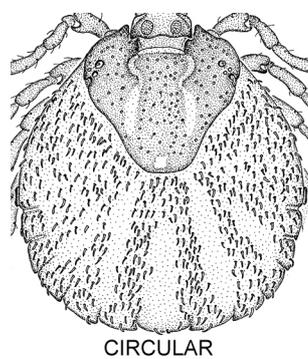
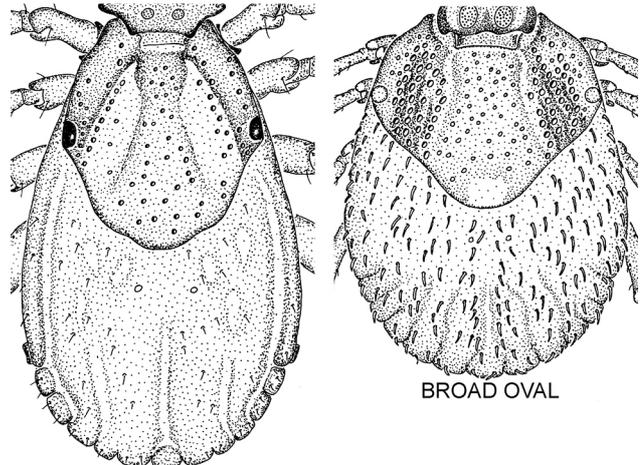


Basis capituli, or **basis**: the single structure to which the sensory and piercing components of the mouthparts are attached. In female ixodid ticks it also bears the porose areas (see below).

Basis dorsal posterior margin: this profile may either be **straight**, or **undulating**.

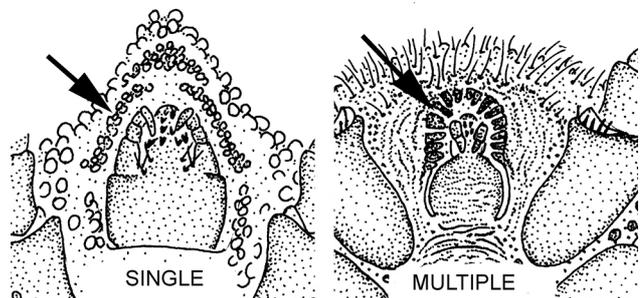


Body profile: in unfed ticks viewed from above the outline of the main body may be a **narrow oval**, or a **broad oval**, or **circular**.



Camerostome: a depression forming a hood-like structure on the anterior ventral surface of argasid ticks, in which the mouthparts are situated.

Camerostome cheeks: the camerostome has cheeks, like flaps, at either side of the mouthparts. On each side they may consist of a **single flap**, or **multiple flaps**.

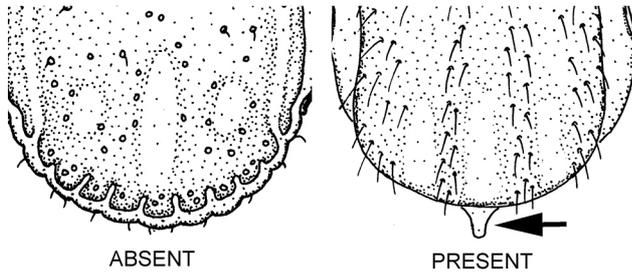


Capitulum: (see Mouthparts) the combined basis capituli and the sensory and piercing mouthparts.

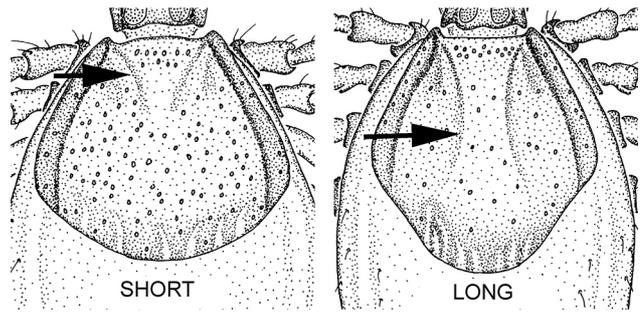
Carinae: see Lateral carinae.

FIGURE 1b. Glossary: Article, to Carinae.

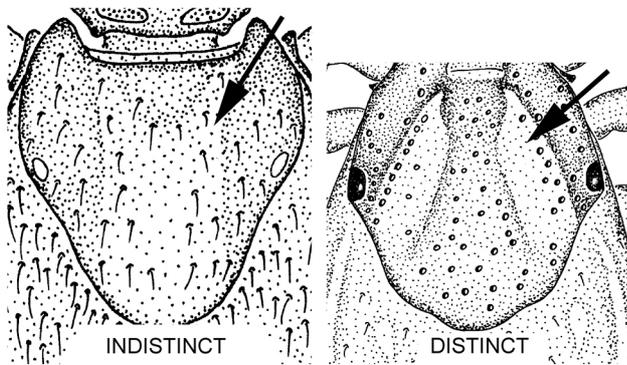
Caudal appendage (of unfed male): in the males of some ticks there is an extension of the body wall like a small tail. This is either **absent**, or **present**. In some species it may also develop when the male feeds and its body expands slightly.



Cervical grooves length: The grooves may be **short**, or **long**.

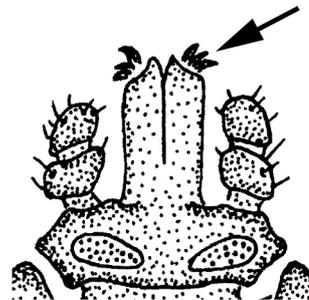


Cervical fields: in female *Rhipicephalus* these are the depressed areas of the scutum between the outer scapular grooves (or lateral carinae) and inner cervical grooves (see Cervical grooves, below). They may be **shallow and indistinct**, or **deep and distinct**.



Cervix: the connection between the capitulum (or gnathosoma) and the idiosoma, like a neck. See also Mouthparts.

Chelicerae: a pair of protrusible rods within the cheliceral sheaths of the mouthparts. At their distal end are teeth cut into the host's skin. The paired cheliceral sheaths are dorsal to the hypostome and form part of the tube for blood sucking and salivation. The cheliceral sheaths are armed with numerous minute teeth on their outer surfaces. (See Hypostome.)



Cervical grooves: (also known as mesial grooves) occur on the scutum or conscutum where it goes down from the central raised area into the depression called the cervical field. These are not true grooves, they are an edge transitional from a high to a low area (also see Lateral carinae).

Columns of teeth on hypostome: the ventral surface of the hypostome has teeth (or denticles) in paired columns. Generally they number 2+2, or 3+3, or 4+4, or 5+5.

Cervical grooves depth: this may be **shallow anteriorly**, or **deep anteriorly**.

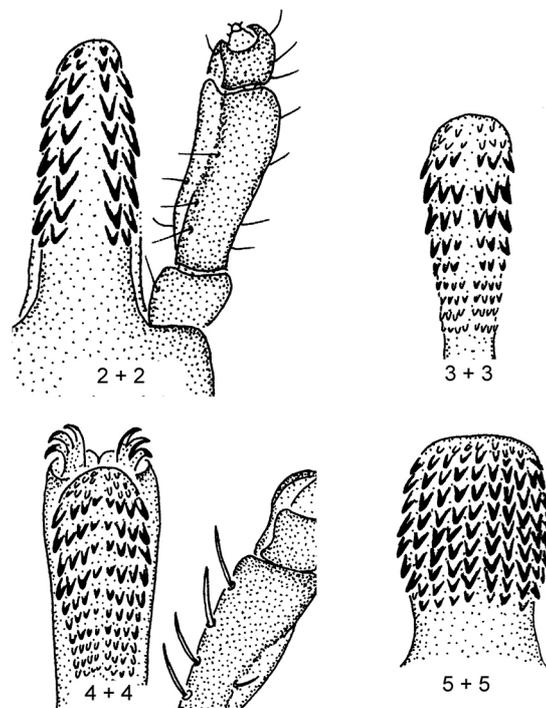
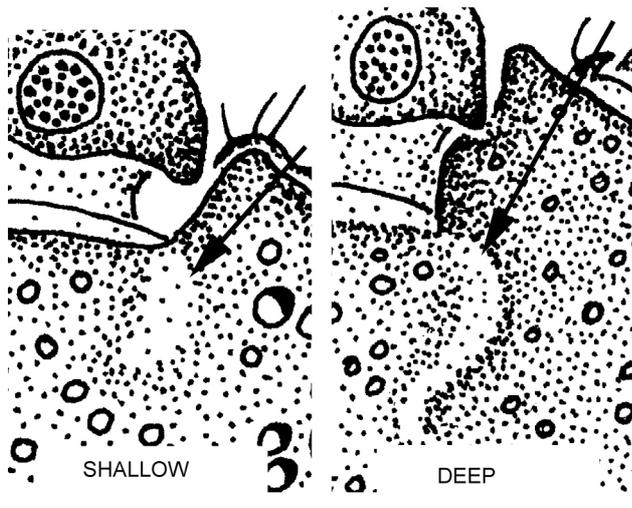
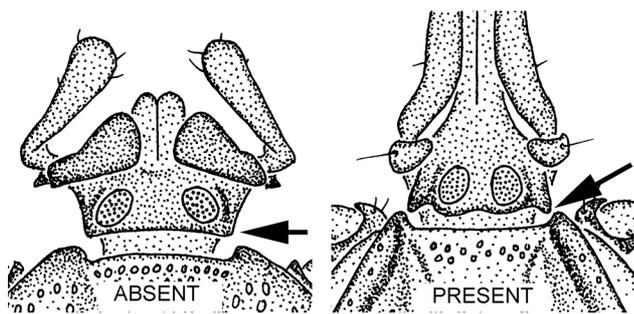


FIGURE 1c. Glossary: Caudal appendage, to Columns of teeth on hypostome.

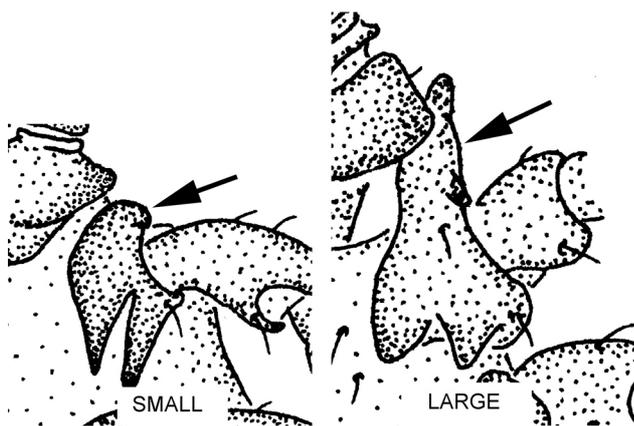
Cornua: the lateral and posterior surfaces of the dorsal basis when they form ridges extending posteriorly. These may be **absent**, or **present**.



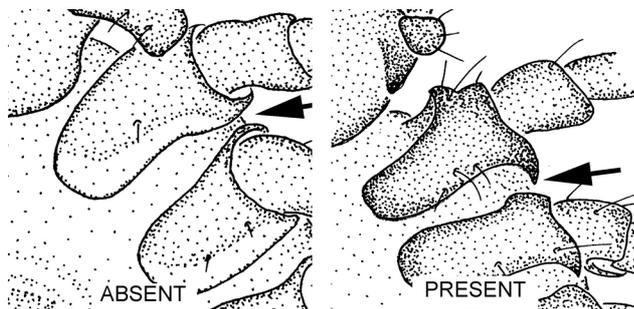
Conscutum (see Scutum): the hard plate covering most of the dorsal surface of male ixodid ticks.

Coxa: the segment of each leg that is fixed to the ventral body wall. Coxae are numbered with the articulating leg, 1 to 4 from anterior to posterior. Coxa 1 may have two, one or no spurs projecting posteriorly.

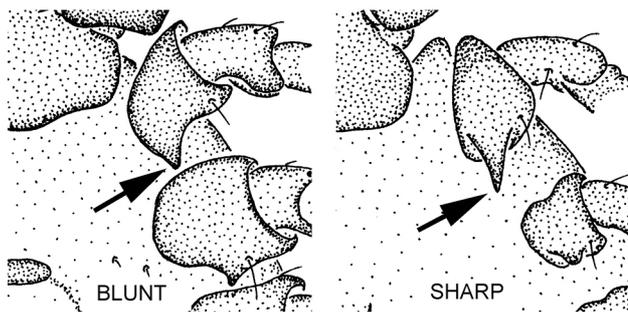
Coxa 1 anterior projection: the first coxa may have a projection anteriorly. This is **small and not visible dorsally**, or **large and visible dorsally**.



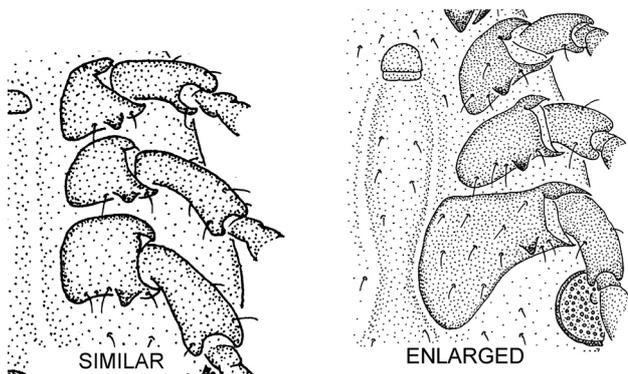
Coxa 1 external spur: the external spur (see Coxa above) may be **absent**, or **present**.



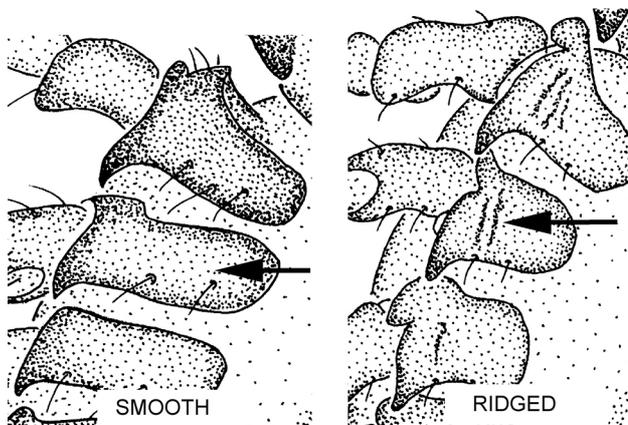
Coxa 1 internal spur: the internal spur (see Coxa above) may be **bluntly pointed**, or **sharply pointed**.



Coxa 4 size (in males): coxa 4 may be of **similar size** to coxae 1 to 3, or it may be **enlarged considerably**.



Coxae surface texture: the surface of the coxa of some *Ixodes* species may be **smooth**, or **ridged**.



Coxae type: the coxae of some *Ixodes* species may have two contrasting textures or colours in their anterior and posterior parts, these are **syncoxae**. Coxae may be **without syncoxae**, or **with syncoxae**.

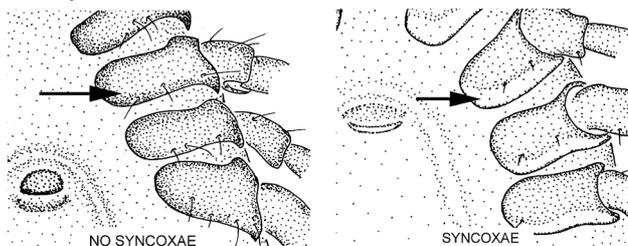
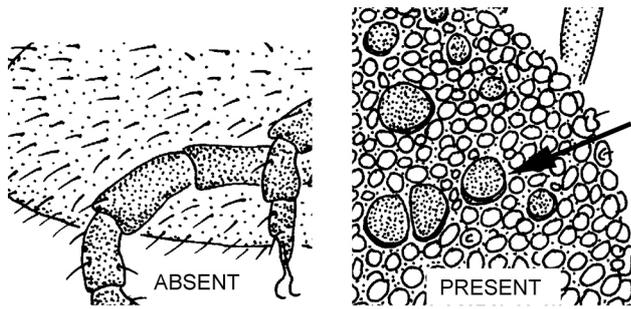


FIGURE 1d. Glossary: Cornua, to Coxa type.

Discs on main body: these are large flattened areas with a distinct round or oval margin, and mainly on the dorsal main body of some species of argasid ticks. They are **absent**, or **present**. (Compare with Mammillae.)

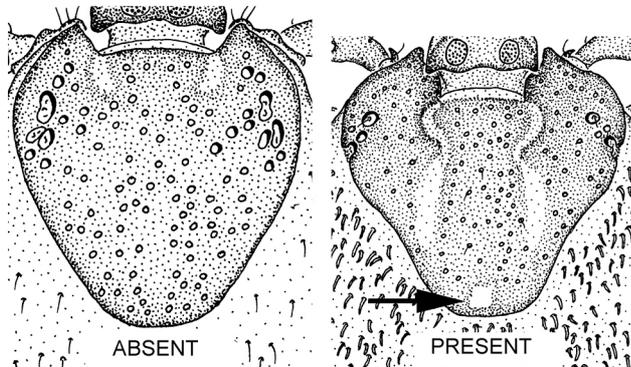


Ditropic: (compare with Monotropic and Telotropic) a type of life-cycle where the immature stages have a feeding preference for a different type of host than the adults.

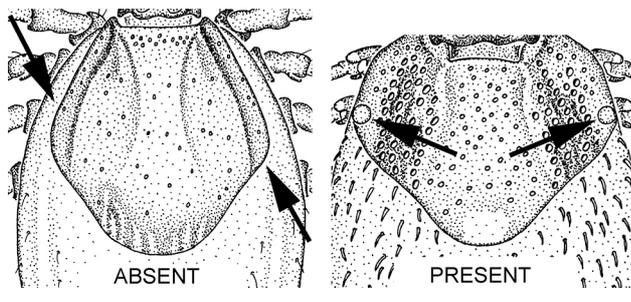
Dorsal: the area of the tick that faces away from the ground or other surface on which the tick is standing with its legs.

Enamel (also known as ornamentation): the integument of some ticks may contain pigment in typical regular patterns, usually on scutum and conscutum. Colours of the pigment include white and yellow through to red. This colour appears like paint, in contrast to the pale bands or whole segments of the legs of some ticks. (See photographs of *Amblyomma* and *Dermacentor*.)

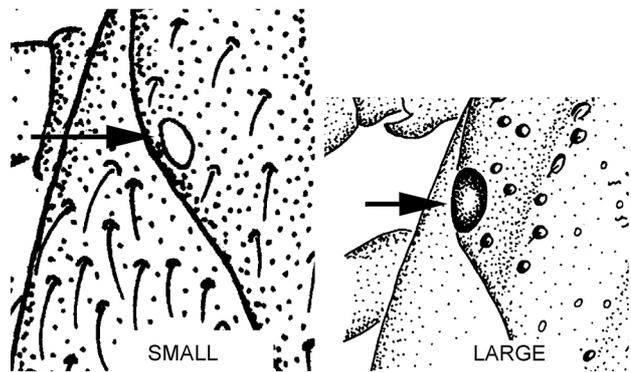
Enamel pattern on scutum or conscutum: this may be **absent**, or **present** (as a single spot or complex pattern).



Eyes: ticks often have parts of the integument modified as light receptors. In Ixodidae eyes occur as a pair at the edge of scutum or conscutum. Some species in the Argasidae have ventral eyes. Eyes are **absent**, or **present**.

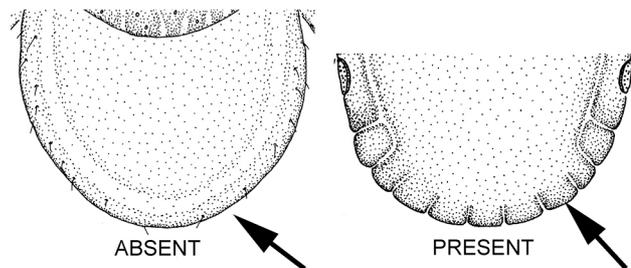


Eye size and profile: this may be **small and flat**, or **large and convex**.

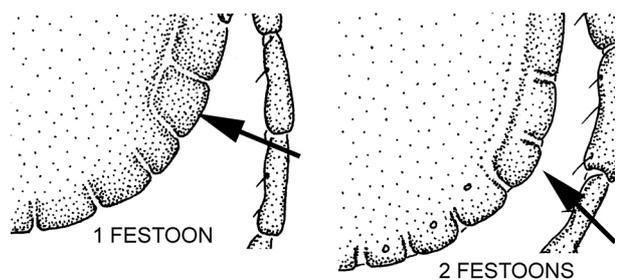


Family: the taxonomic group at the level above genus. For example Argasidae is a family that contains genera such as *Argas*, *Ornithodoros* and *Otobius*.

Festoons: the posterior margin of the body of many female and male ixodid ticks forms into regular folds and grooves called festoons (they disappear in engorged ticks). They may be **absent**, or **present**.



Festoons of female enclosed by lateral groove: the lateral groove may enclose 1 festoon, or 2 festoons.



Festoons of male enclosed by lateral groove: the lateral groove may enclose 0 festoons, or 1 festoon.

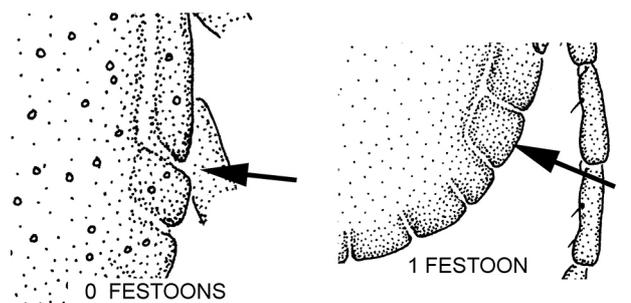
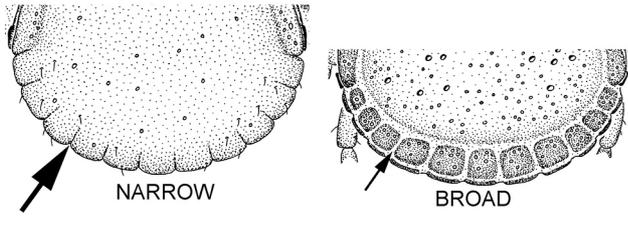
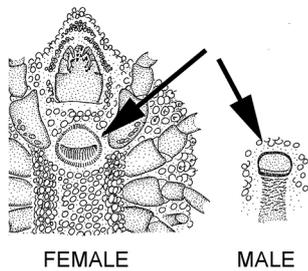


FIGURE 1e. Glossary: Discs on main body, to Festoons of male enclosed by lateral groove.

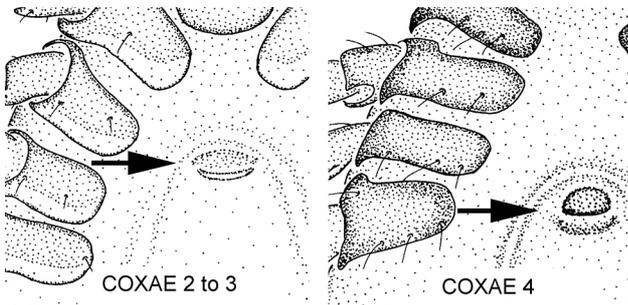
Festoon grooves: the grooves which separate individual festoons of males, both dorsally and ventrally, may be **narrow**, or **broad**.



Genital aperture: in adult ticks this is a large aperture situated between the coxae. The genital aperture in argasid ticks is similar to that in ixodid ticks but the male aperture is smaller than the female (figure below is *Ornithodoros capensis* female ventral). In ixodid ticks the apertures of females and males are similar in size but male apertures are covered by an apron.



Genital aperture position: this may be **level with coxae 2 to 3**, or **level with coxae 4**.

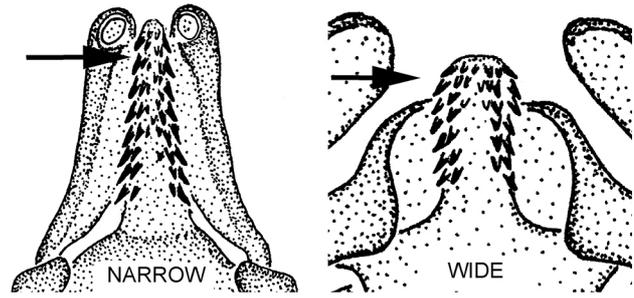


Genus: the taxonomic group above the level of species. The argasid tick genus *Otobius* comprises just three species, *O. megnini*, *O. lagophilus* and *O. sparmus*.

Hypostome: the ventral section of the passage for blood sucking and salivation. Its outer surface has teeth for attachment.



Hypostome shape: in *Ixodes* species this may be **narrow at anterior**, or **wide at anterior**.



Instar: one of the stages of the tick life-cycle. The egg, larva, nymph and adult are the four separate instars of ixodid ticks. The argasid ticks have up to 6 or more nymphal instars, called first, second etc.

Integument: this is the outer body wall of ticks, formed by the inner living epidermis and the outer cuticle. It forms an exoskeleton for attachment of muscles, and a tough protective surface. The cuticle consists mainly of protein and chitin, an outer layer of wax for waterproofing. In areas of the tick that are not sclerotized the integument is flexible at joints, or may grow and stretch to accommodate the blood meal.

Ixodidae: within the group Acari the ticks are in the group Ixodida and within this there are two main families, the Ixodidae and the Argasidae. Ixodid ticks are also known as the hard ticks because they have hardened (sclerotized) plates on their surface.

Lateral carinae on scutum: in some female *Ixodes* ticks these are a ridge on the outer margin of the cervical fields and start near the scapulae. (Also known as scapular grooves in female *Rhipicephalus*. Not to be confused with Lateral grooves, below. These carinae are not true grooves, they are an edge transitional from a high to a low area.) **Lateral carinae are absent**, or **present**.

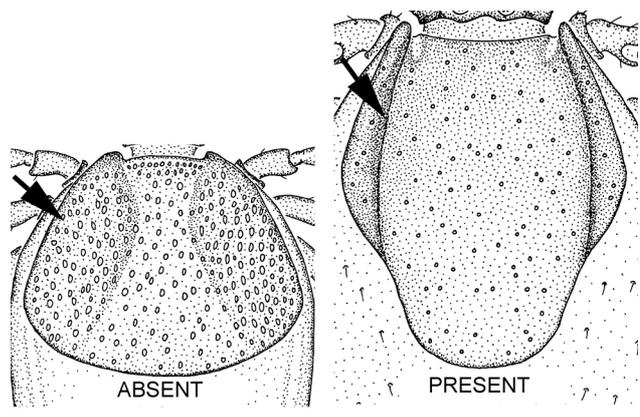
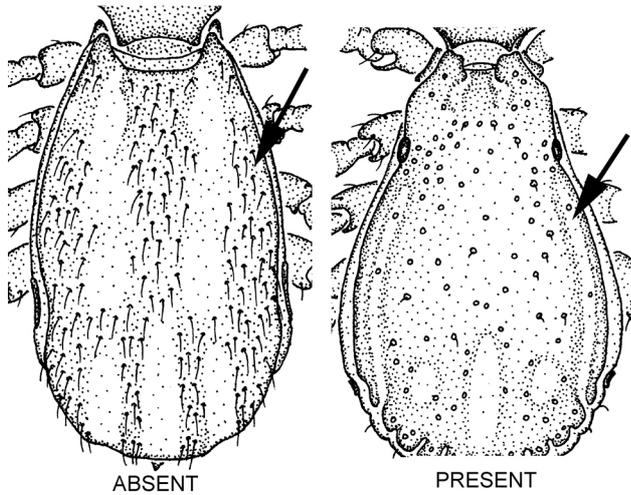
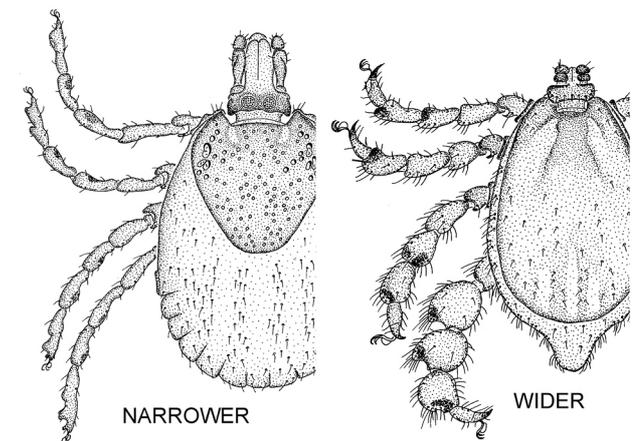


FIGURE 1f. Glossary: Festoon grooves, to Lateral carinae on scutum.

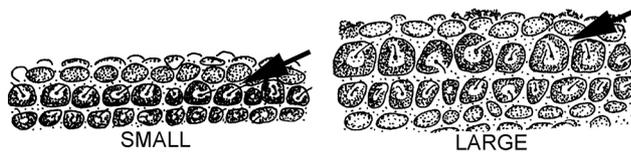
Lateral grooves: (also known as marginal grooves or lines) these are grooves at the side margins of the conscutum of male ticks. They may be **absent**, or **present**.



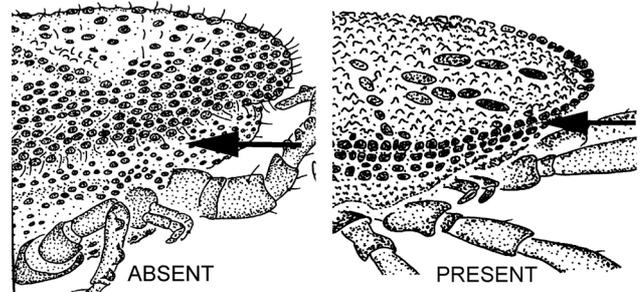
Leg width: this is in general of the **same width as the mouthparts**, or in (especially in males) **wider than the mouthparts**.



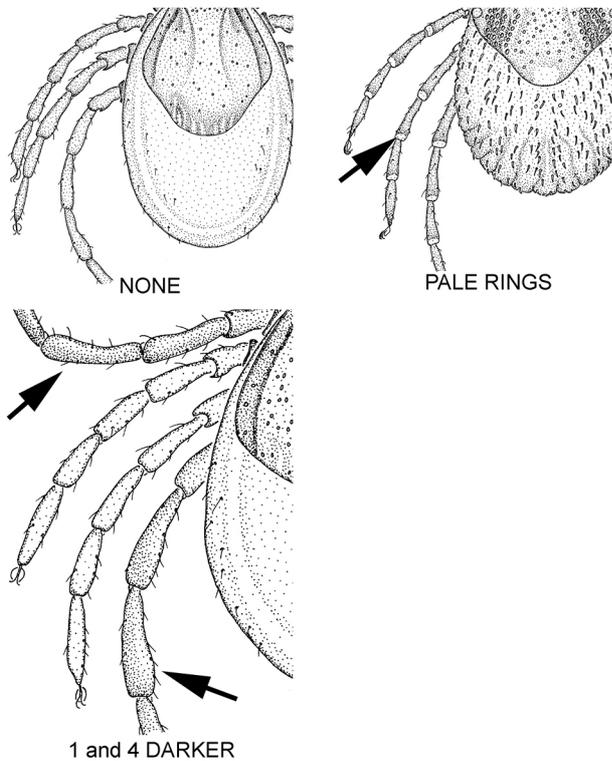
Lateral suture mammillae: *Argas* ticks have a clear margin between dorsal and ventral surface, known as a suture. Along the suture the dorsal mammillae may be **small**, or **large**.



Margin between dorsal and ventral surface: in some argasid ticks a distinct division line between dorsal and ventral surfaces is **absent**, in other argasid ticks a distinct division (known as a lateral suture) is **present**.



Leg colour pattern: some ticks have different depths of pale to dark brown colour of the segments of their legs. Colour patterns may be **absent**, or **with pale rings** at distal end of segments, or **legs 1 and 4 darker than legs 2 and 3**.



Marginal groove: this is a feature of the conscutum of male *Amblyomma* and *Bothriocroton* ticks (not the same structure as the lateral groove of many *Rhipicephalus* ticks). It may be formed by a line of punctations continuing to first festoon only, or a shallow groove with irregular ridges continuing around anterior margin of all festoons, or a deep and smooth groove continuing around anterior edge of all festoons.

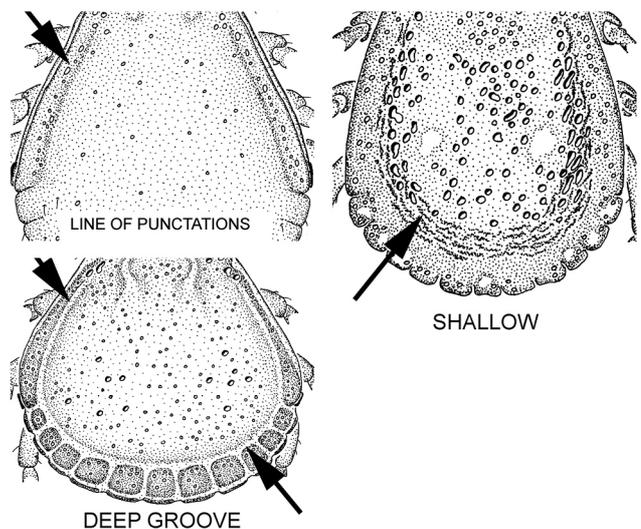
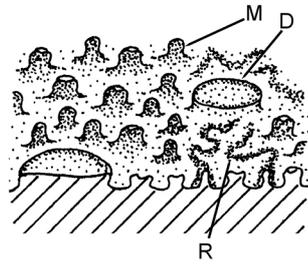
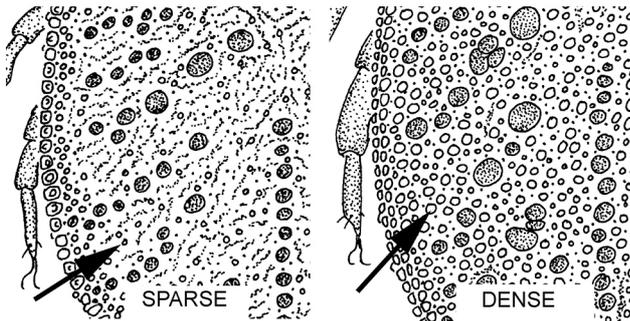


FIGURE 1g. Glossary: Lateral grooves, to Marginal groove.

Mammillae (also known as buttons): the integument of many argasid ticks have complex protrusions; one type has raised, rounded humps called Mammillae (M). Other protrusions are sharp Ridges (R) in complex patterns, and flat-topped Discs (D) in a radial pattern.



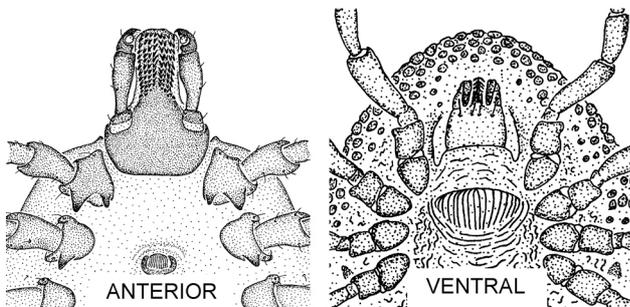
Mammillae distribution: may be sparse, or dense.



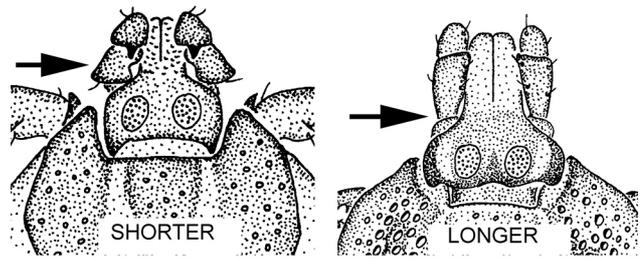
Monotropic: (compare with Ditropic and Telotropic) a type of life-cycle where the immature stages and adults share feeding preference for the same type of host, such as *Argas persicus* feeding on birds. (This is different from a 1-host life-cycle, where a single tick feeds as larva then nymph then adult in one sequence on a single individual of a species of host.)

Mouthparts: comprise the palps, hypostome, cheliceral sheaths and chelicerae. All these parts are mounted on the basis capituli to form a feeding apparatus. The palps do not pierce the host during feeding, whilst the hypostome and cheliceral sheaths form the piercing and blood sucking tube. In ticks and mites the fundamental segmentation of arthropods is reduced so that the body consists of two parts, the gnathosoma (or capitulum) at the anterior comprising the mouthparts and basis, and the posterior idiosoma containing the internal organs. The gnathosoma can articulate on the idiosoma during feeding and egg laying.

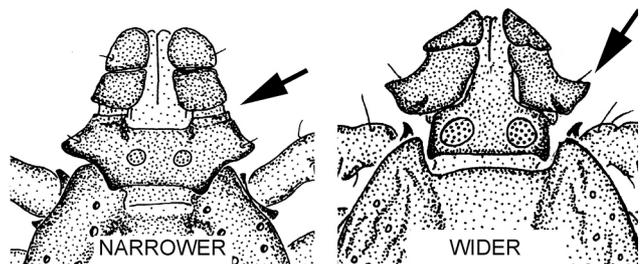
Mouthparts position: they either project from the body outline anteriorly, or they are entirely ventral.



Mouthparts length: they are either the same or shorter than the basis, or longer than the basis.

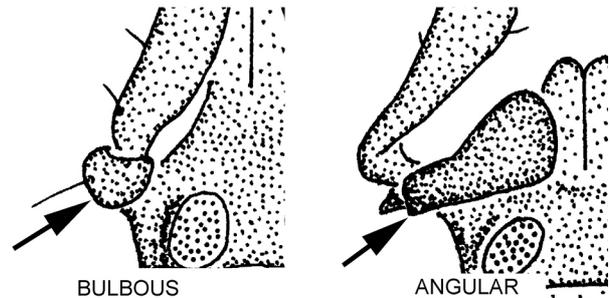


Mouthparts width: they are either the same or narrower than the basis, or wider than the basis.



Palps: (see also Article) a pair of segmented structures either side of the central tube-like part of the mouthparts. In ixodid ticks an important function is detecting a suitable site for the tick to feed; they also splay apart on the surface of the host's skin during feeding of ixodids and may assist in spreading attachment cement to the skin.

Palp article 1: this may be shaped as bulbous and distant from cheliceral sheath, or widely angular and extends over cheliceral sheath.



Palp article 2 external margin: the ventral side may be slightly concave, or distinctly concave.

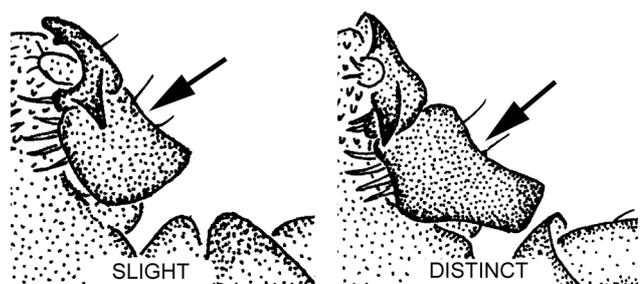
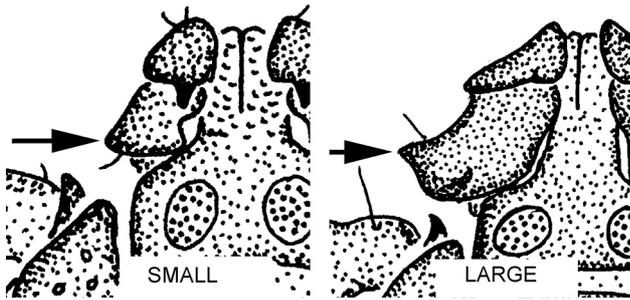
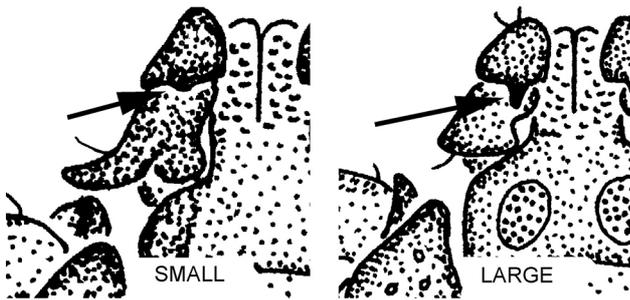


FIGURE 1h. Glossary: Mammillae, to Palp article 2 lateral margin.

Palp article 2 lateral projection: this may be **small**, or **large**.



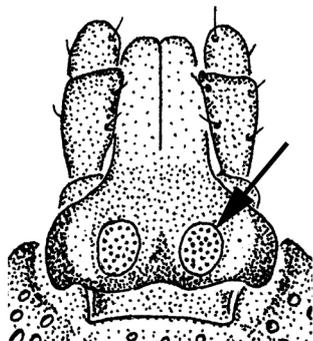
Palp article 3 dorsal posterior spur of male: this may be **small**, or **large**.



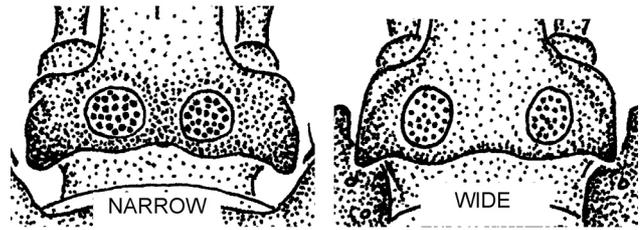
Palps shape: viewed dorsally or ventrally this has a general appearance as **long and slender**, or **short and thick**.



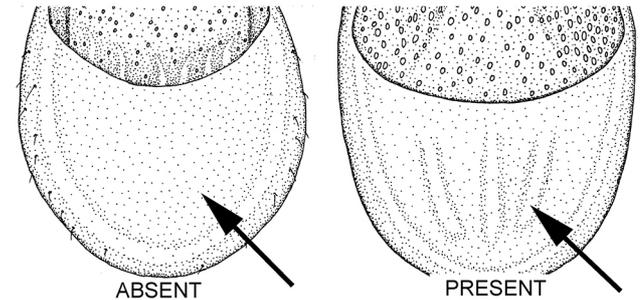
Porose areas: the dorsal surface of the basis of female ixodid ticks has two patches of fine pores. Secretions through the pores waterproof the eggs as they are laid and manipulated by a special structure called Gené's organ that extends out from the cervix. (The cervix is the articulation between the basis and the rest of the body.)



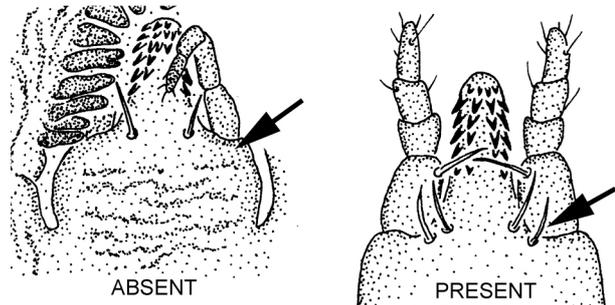
Porose areas separation: the gap between the porose areas may be **narrow**, or **wide**.



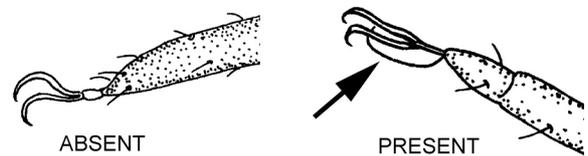
Posterior grooves (of unfed females): depressions in the surface of the alloscutum of some ixodid ticks may be **absent**, or **present**.



Postpalpal setae (see below for Setae): in argasid ticks there is a set of distinct setae on the ventral surface of the basis, with a pair posterior to the palps may be **absent**, or **present**.



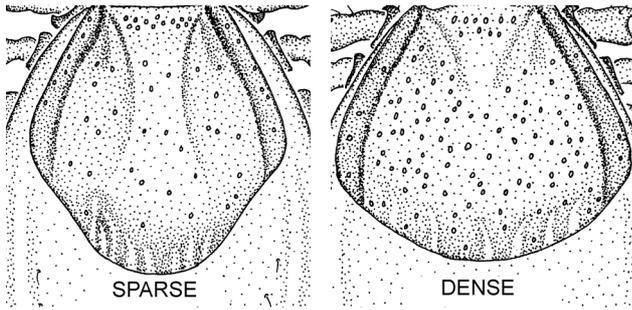
Pulvilli: these are soft white pads between the gripping claws at the end of the tarsi. They grip onto smooth surfaces. Pulvilli are **absent** in the Argasidae ticks, but are **present** in the Ixodidae ticks.



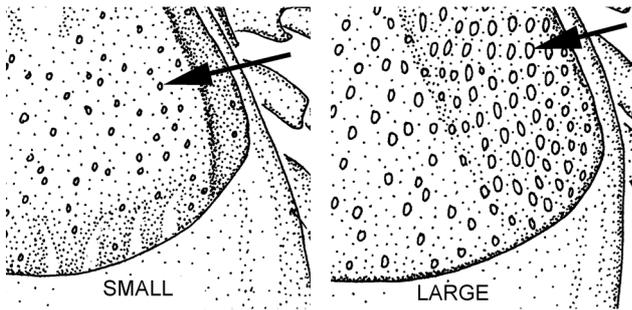
Punctuation: punctations are pits in the surface of the scutum, conscutum and other sclerites. They are openings of secretory glands or contain sensory setae. Their size varies greatly between species and the smallest are not readily visible by light microscopy.

FIGURE 1i. Glossary: Palp article 2 lateral projection, to Punctuation.

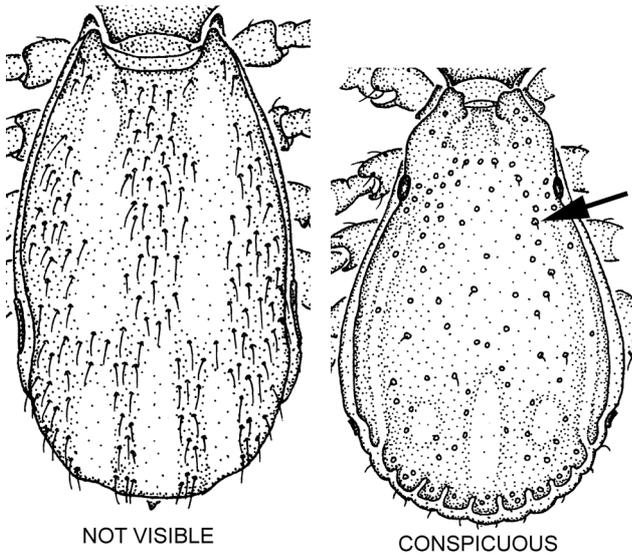
Punctuation distribution on scutum or conscutum: these generally may be **sparse**, or **dense**.



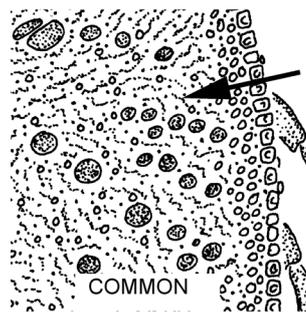
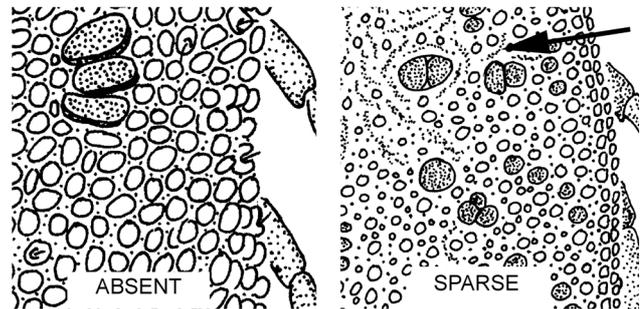
Punctuation size on scutum or conscutum: the individual punctations may be **small**, or **large**.



Punctations on conscutum or scutum: may be **not visible**, or **conspicuous**.

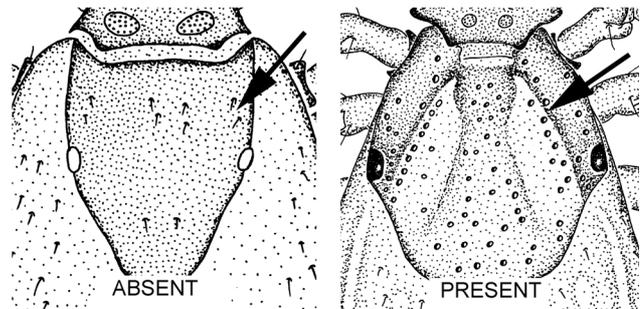


Ridged areas on main body: these are linear raised areas of the integument that form striated or networked patterns on many species of argasid ticks. They may be **absent**, or **sparse**, or **common**.



Scapulae: the pair of most anterior points of the scutum or conscutum at either side of the basis, like shoulders.

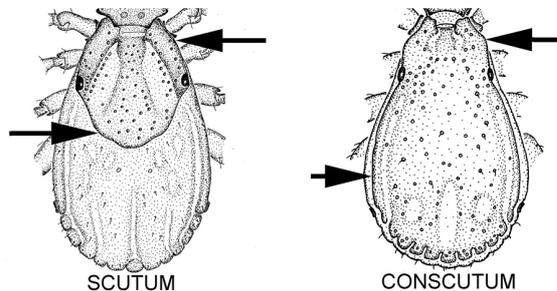
Scapular grooves of female: (also known as Lateral carinae in *Ixodes* and not to be confused with Lateral grooves, above). In some *Rhipicephalus* ticks these are formed as a ridge on the outer margin of the cervical fields and start near the scapulae. (These are not true grooves, they are an edge transitional from a high to a low area.) They are **absent**, or **present**.



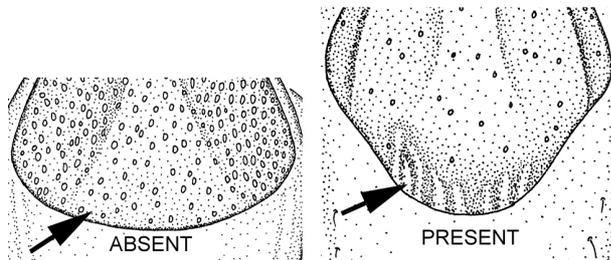
Sclerotize: a hardening of the integument to stiffen it. Sclerotin is a protein that can be variably hardened by quinone tanning to enable the leg segments to act as an exoskeleton, or to harden flat structures on the body wall such as the scutum, anal plates and ventral plaques. (These structures are known generally as sclerites.)

FIGURE 1j. Glossary: Punctuation distribution on scutum, to Sclerotize.

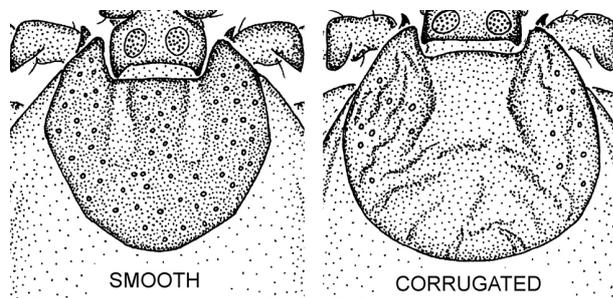
Scutum and conscutum (compare with Alloscutum): the scutum is a large hard plate on the dorsal anterior surface of the body of female ixodid ticks. The conscutum is a similar plate on males that covers most of the dorsal surface. The integument of the posterior body of female ixodids is not sclerotized so that the females can expand greatly during feeding and this area is called the alloscutum.



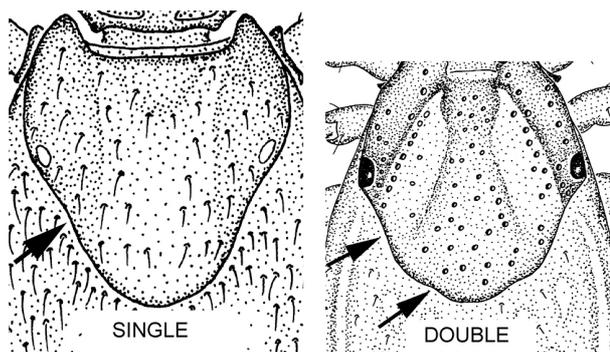
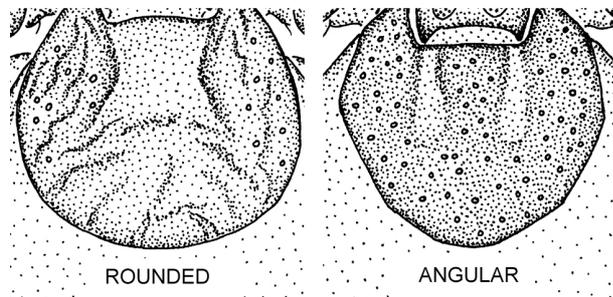
Scutal corrugations at posterior: the scutum close to the posterior margin may have a series of small grooves forming a corrugated area. This may be **absent**, or **present**.



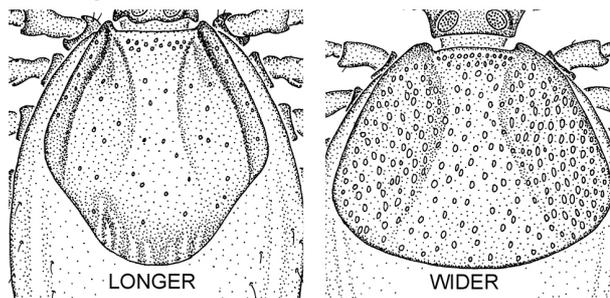
Scutum general texture: this may be **smooth**, or **corrugated**.



Scutum posterior margin: on females this may be **smoothly rounded**, or **slightly angular**; or have a **single concavity each side**, or **double concavity each side**.

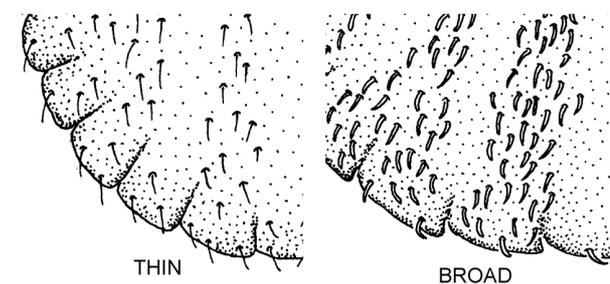


Scutum proportions: these may be **longer than wide**, or **wider than long**.



Setae (also called hairs) are long thin extensions of the integument. Setae occur as various types; simple, branching at the tip (pectinate), or as spines. (In zoology, hair is a diagnostic character of mammals.)

Setae on alloscutum only: these may be **thin and colourless and sparse**, or **broad and white and numerous**.



Setae on both scutum and alloscutum: these may be **short and sparse**, or **long and numerous**.

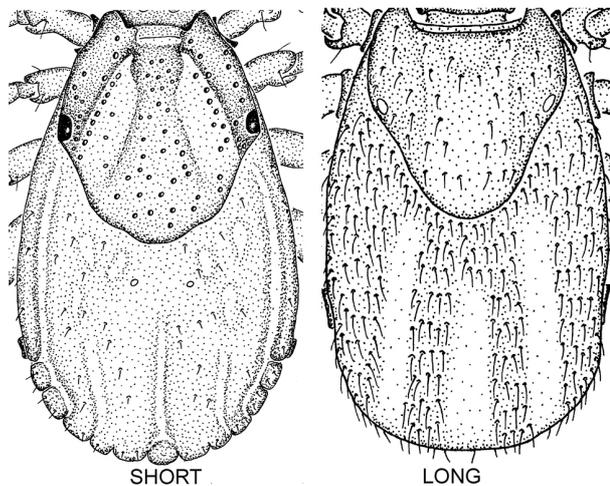
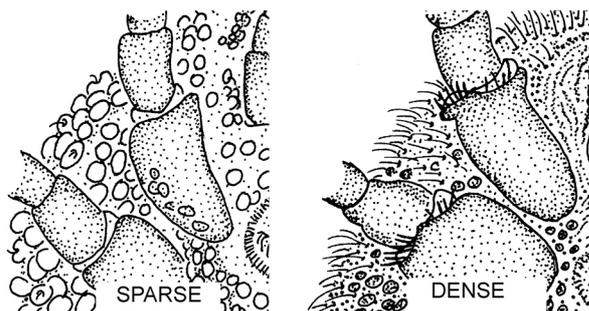


FIGURE 1k. Glossary: Scutum and conscutum, to Setae on both scutum and alloscutum.

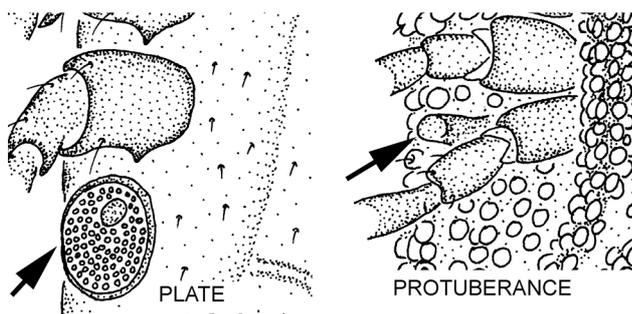
Setal density on ventral surface: these may be distributed in a pattern that is **sparse**, or **dense**.



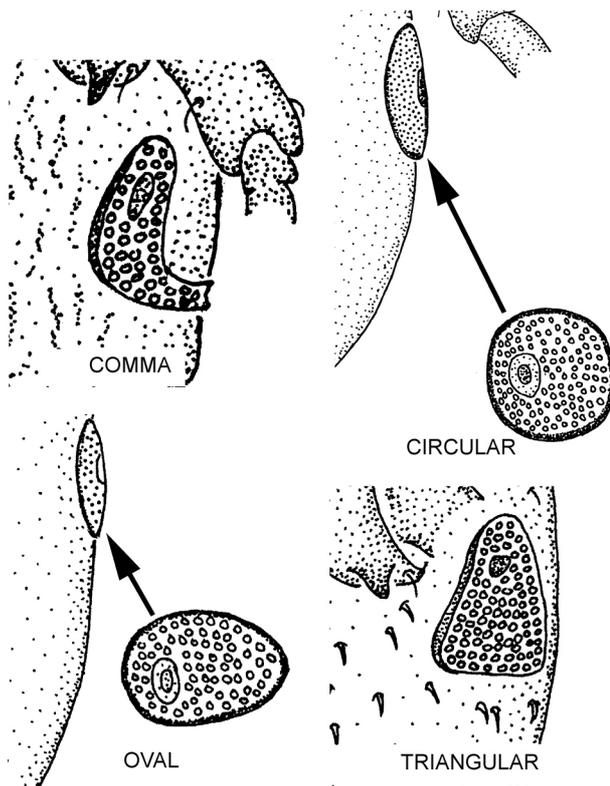
Species: for ticks a species is a population in which individuals are all capable of interbreeding to produce fertile offspring of the same kind. In practice it is difficult to define the term species, for example some species may interbreed sufficiently to produce fertile offspring. Species have a binomial (or binomial) name consisting of the genus (for example *Ixodes*) followed by the specific name (for example *holocyclus*). This may be followed by the name of the person who first described the species, plus that date. Species names are always in italics. Sometimes a trinomial name is used in cases of difficulty with differentiating closely similar species. The concept of species is one of the fundamental problems of biology. Readers of this publication should be cautious when they use species names, and should expect that some of the names used here will change because of new information, particularly from studies of tick's genetic material.

Spines: (compare with Setae) these are a type of setae covering the general body wall, they are rigid, thick at the base and sharply tapering. They are characteristic of *Otobius* ticks (see main drawing).

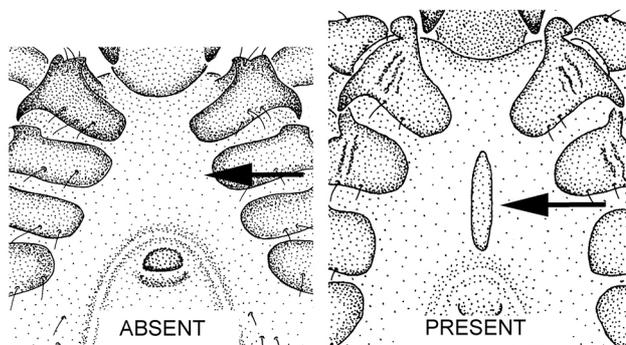
Spiracle opening: ticks respire through a system of internal air tubes known as tracheae. The external opening is a slit on the spiracle plate. Ixodid ticks have this opening on a large sclerotised plate that also bears many hollow spaces known as goblets. In ixodid ticks there is a **large spiracle plate posterior to legs 4**. Argasid ticks have only a simple opening, without the plate and on a **protuberance between legs 3 and 4**.



Spiracle plate shape: this may be like **comma**, or **circular**, or **oval**, or **triangular**.



Sternal plate: this is a single oblong sclerotised area on the ventral surface of some *Ixodes* females. It is either **absent**, or **present**.



Tarsi: these are the distal or outermost segments of the legs of ticks.

Tarsi distal profile: in *Ixodes* species this is either **gradually stepped**, or **steeply stepped**.

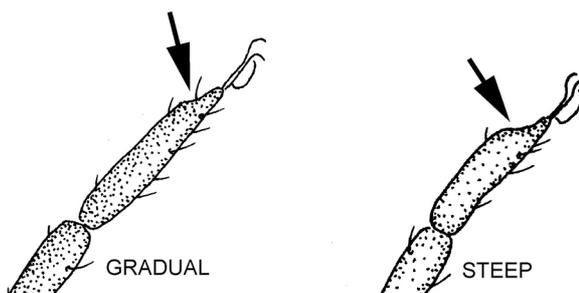
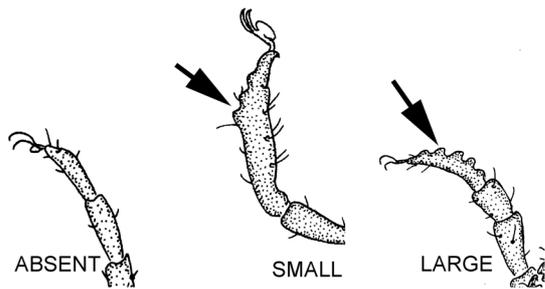
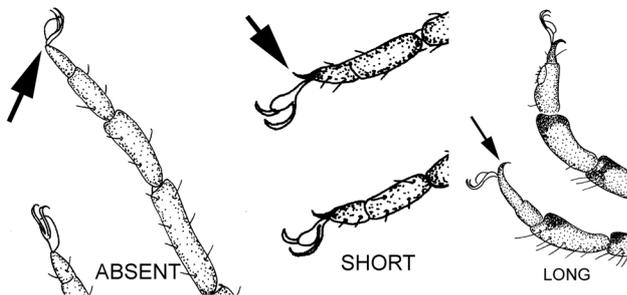


FIGURE 11. Glossary: Setal density on ventral surface, to Tarsi distal profile.

Tarsal dorsal humps: these occur as a series of sharp undulations and are **absent**, or **small**, or **large**.

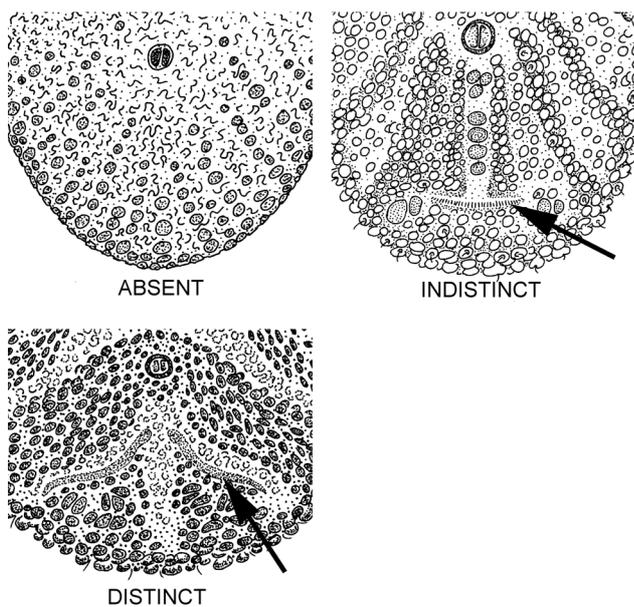


Tarsal terminal spurs: these are single extensions of the end of each leg that act like a claw, in addition to the paired claws of ixodid ticks. Relative to the paired claws they may be **absent**, or **short**, or **long**.

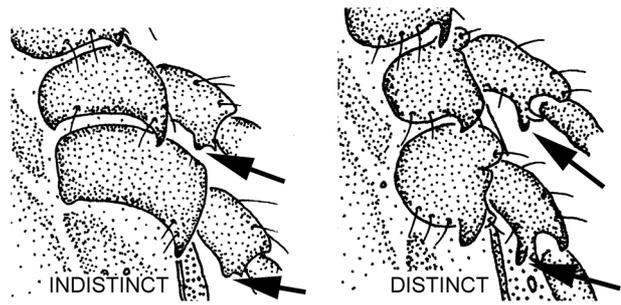


Telotropic: (compare Monotropic and Diptropic) a type of life-cycle where the immature stages have feeding preferences for both a different type of host and the same type of host as the adults.

Transverse postanal groove: (also known as paired organ) in argasid ticks this is a fold in the posterior ventral integument with a different texture and may be **absent**, or **indistinct and single**, or **distinct and paired**.

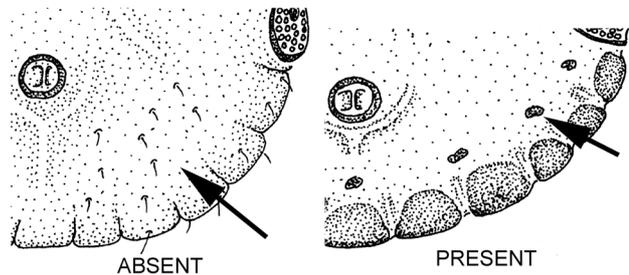


Trochanter spurs: the trochanters are the first moveable segment of the legs, beyond the coxae. Ventrally on legs 3 and 4 they may have a posterior spur that is **indistinct** or **distinct**.



Ventral: this is the area of the tick that faces toward the ground or other surface that the tick is standing on with its legs.

Ventral plaques: small sclerotized patches on the posterior ventral surface of males of some species of ticks. They are **absent**, or **present**.

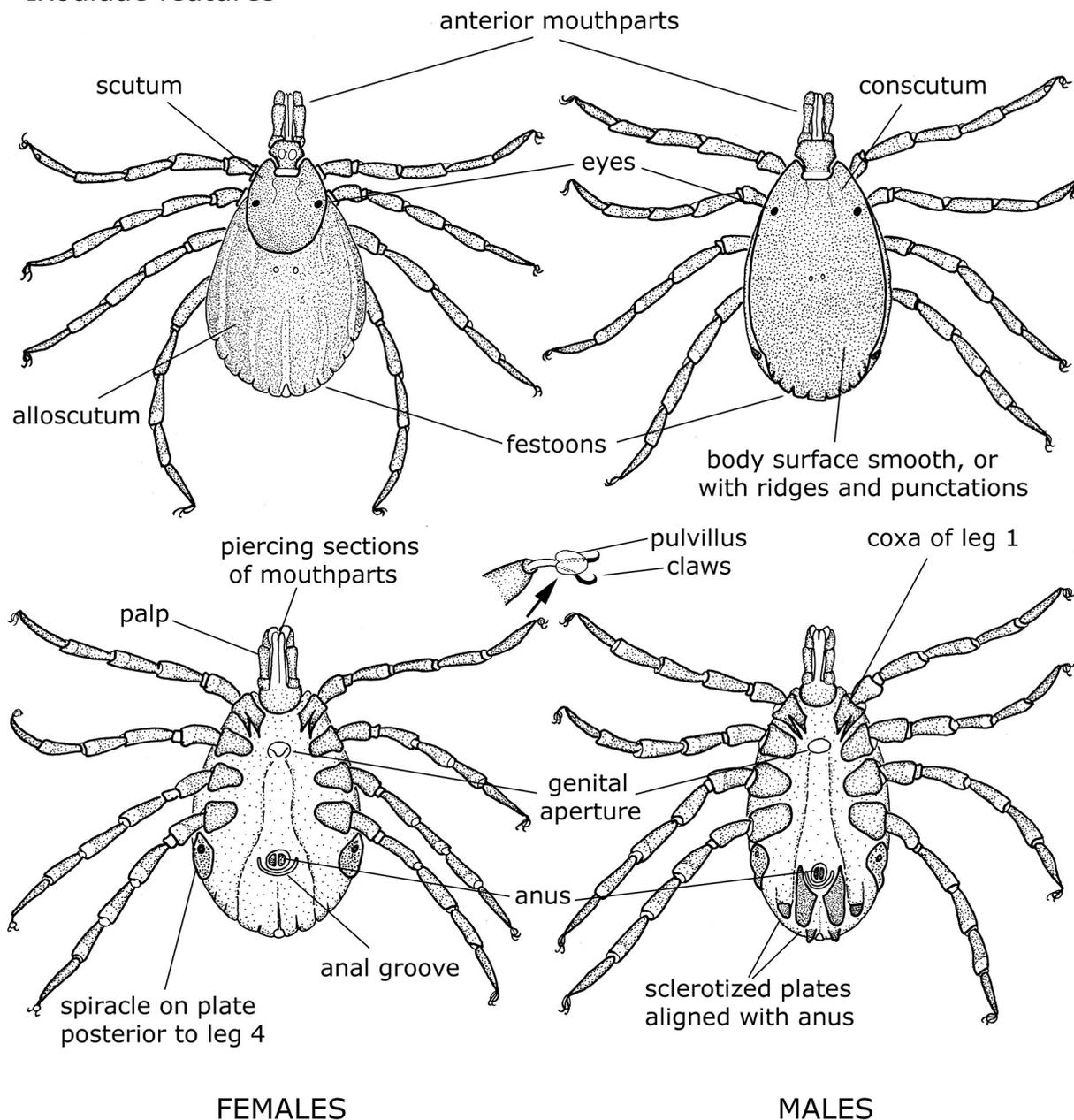


Ventral plates: a term sometimes used for Anal plates (not to be confused with ventral plaques).

FIGURE 1m. Glossary: Tarsal dorsal humps, to Ventral plates.

Australia. We speculate that the Nutalliellinae may well have lived in Australia (Barker, Walker & Campelo in prep.), or might still be here, but certainly the Rhipicephalinae evolved elsewhere (probably in Africa) and much, much later (Murrell *et al.* 2001a). Whereas the Australian tick fauna is rich in subfamilies, it is depauperate in species: only 65 of the 896 species of ticks are endemic to Australia (Barker, Walker & Campelo in prep.). Furthermore, the earliest diverging lineage of the subfamily Ixodinae, the Australasian *Ixodes*, is endemic to Australasia (Barker & Murrell 2004). The closest living relatives to the ticks, the sister-group, seems to be the free-living holothyrid mites (Murrell *et al.* 2005 and references therein) which also occur in Australia.

Ixodidae features



FEMALES

MALES

FIGURE 3. Morphological features of hard ticks (family Ixodidae). Example is an adult female and an adult male of the genus *Hyalomma*. Top row is dorsal view, bottom row is ventral view.

Feeding of ticks in relation to animal and human health

It is the feeding of ticks that makes them important to the health of domestic animals and humans. During feeding, ticks cause harm to their hosts by taking blood, injuring the skin, causing irritation and pain, and sometimes by

causing poisoning. More serious disease is often caused when the ticks are infected with microorganisms. These are transmitted to humans or domestic animal when the ticks feed and may cause weakening or fatal disease in the host. This type of microorganism is known as a pathogen and when a tick transmits it to a host the tick is known as a vector of the pathogenic microorganism. Strictly speaking, ticks do not transmit diseases, they transmit pathogens which may cause the clinical conditions defined as a particular disease.

Ticks feed only on the blood (blood cells and blood plasma) and lymph of their hosts. The ticks crawl onto their host and attach to the skin. Ticks use a combination of cutting mouthparts, which penetrate the skin, and often an adhesive (cement) is secreted in the saliva to aid attachment to the skin (Fig. 4). At the end of the mouthparts are sharp chelicerae that scrape a hole into the dermis. This breaks the capillary blood vessels very close to the surface of the skin and the tick feeds on released blood and lymph which may accumulate at the wound. On the ventral surface of the mouthparts is the hypostome which is barbed with teeth to grip the host. A feeding tube into the tick is formed loosely between the hypostome and the sheath surrounding the chelicerae.

The feeding of ixodid ticks is slow because the body wall needs to grow before it can expand to take a very large blood meal. Larvae take 3 to 5 days to fully engorge with blood, nymphs 4 to 8 days, and females 5 to 20 days. When the ticks have engorged with blood they detach from the host's skin and drop to the ground. Males of most species of ticks feed but do not expand like the females. They feed enough for their reproductive organs to mature. Male ticks in the genus *Ixodes* have active reproductive organs as soon as they moult from the nymphal stage and usually do not feed. The argasid ticks usually feed more rapidly, in minutes to several hours. They only take small blood meals but take several of them in each stage of the life-cycle (except for a few argasid species with non-feeding larvae). Argasid ticks do not have the complex attachment to the skin that ixodid ticks have.

Host finding

All ticks spend most of their life-cycle away from their hosts, hiding either in soil and vegetation or in the nests of their hosts. So they need to be able to find hosts on which to feed. Ticks do this in several ways. The eggs and moulting stages of many ticks are in soil or vegetation in the environment in which their hosts graze or hunt. The ticks crawl onto vegetation and wait for their hosts to pass by. This is a type of ambush and the behaviour of waiting on vegetation is called questing (Fig. 4). Thus, in three-host ixodid ticks, like most of the *Rhipicephalus* species, the larvae, nymphs and adults quest on vegetation. The ticks grab onto the hosts using their front legs and then crawl over the skin to find a suitable place to attach and feed.

Adult ticks of the genera *Amblyomma* and *Hyalomma* (the latter does not occur in Australia) are active hunters and will run across the ground to hosts that are resting nearby. Other ticks, such as nearly all argasid (soft) ticks, and many *Ixodes* species, spend their life-cycle in or near the nests or shelters of their hosts (endophilic or nidicolous behaviour). Some ticks are adapted to living in the housing of domestic animals. Attractants released by hosts that stimulate questing by ticks include carbon dioxide and ammonia.

Tick reproduction and life-cycles

Mating. In the hard ticks, mating takes place on the host, except for *Ixodes* species where mating may also occur when the ticks are still on the vegetation or in the nest of the host. Male ticks may remain on the host and will attempt to mate with many females between feedings. The males transfer a sac of sperm (= spermatheca) to the female. The females mate only once, before they are ready to engorge fully (to repletion) with blood. When they are finally engorged, females detach from the host and have enough sperm stored to fertilize all their eggs. Female ixodid ticks lay many eggs (2,000 to 20,000) in a single batch, then they die. Female argasid ticks lay repeated small batches of eggs. Eggs of all ticks are laid in the physical environment, never on the host.

Life-cycle of three-host ticks. Figure 5 shows the sequence of feeding and moulting during the life of individual ticks from a single batch of eggs of a typical three-host species. This is by far the most common type of life-cycle. Larvae develop in the eggs until they are ready to hatch, usually in several weeks. Larvae feed once on a host, then detach from the host and hide in the physical environment such as soil or vegetation. Larvae then moult to nymphs. Nymphs feed once and moult in the same way as larvae. From the nymphal moult, either a female or male emerges. The female feeds once, lays a batch of eggs, then dies. The males may take several small feeds, mate repeatedly, then die. Ticks that have recently hatched from eggs or from moulting have soft bodies and are inactive for one to two weeks whilst the external body wall hardens. The life-cycle of three-host ticks is slow, from six months to several years to complete.

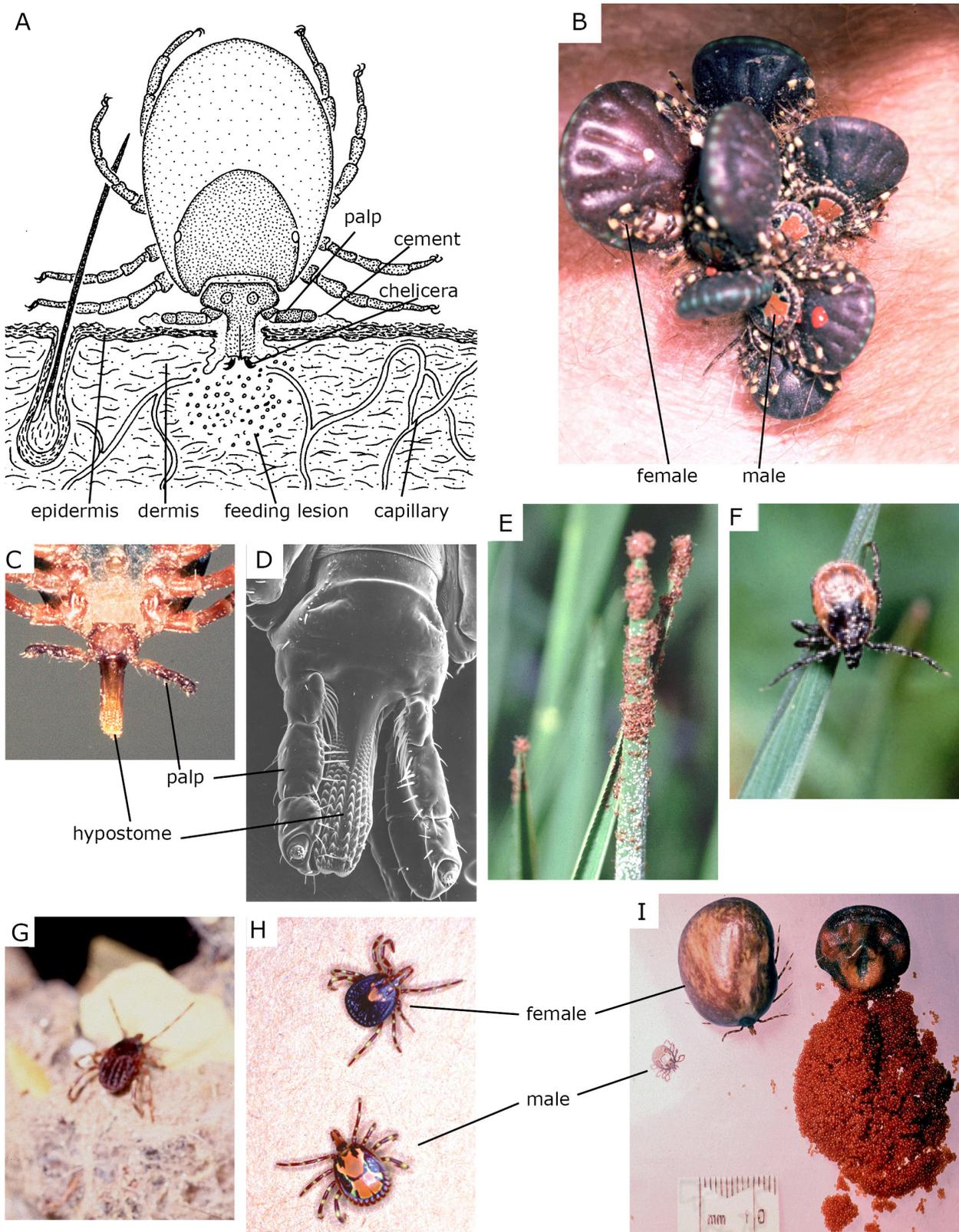


FIGURE 4. Biological characteristics of ticks. (A) Female ixodid tick, feeding position (dorsal surface of tick). (B) Feeding aggregation of ixodid ticks, *Amblyomma variegatum* (the tropical bont tick), on a heifer. (C) Ventral surface of ixodid tick, *Amblyomma variegatum* showing splayed palps in feeding position. (D) Ventral surface of ixodid tick, *Hyalomma anatolicum* (no common name) showing teeth on the hypostome. (E) Larvae of *Rhipicephalus microplus* (cattle tick) questing on grass. (F) Adult female *Ixodes ricinus* (castor bean tick) questing on grass. (G) Adult female *H. anatolicum* actively seeking a host. (H) Adults of *A. variegatum* actively seeking a host. (I) Adults of *A. variegatum*; fed male, engorged female, and exhausted female after laying the batch of approximately 20,000 eggs. [These species of ticks do not occur in Australia but are nonetheless instructive about the biology of the Australian ticks.]

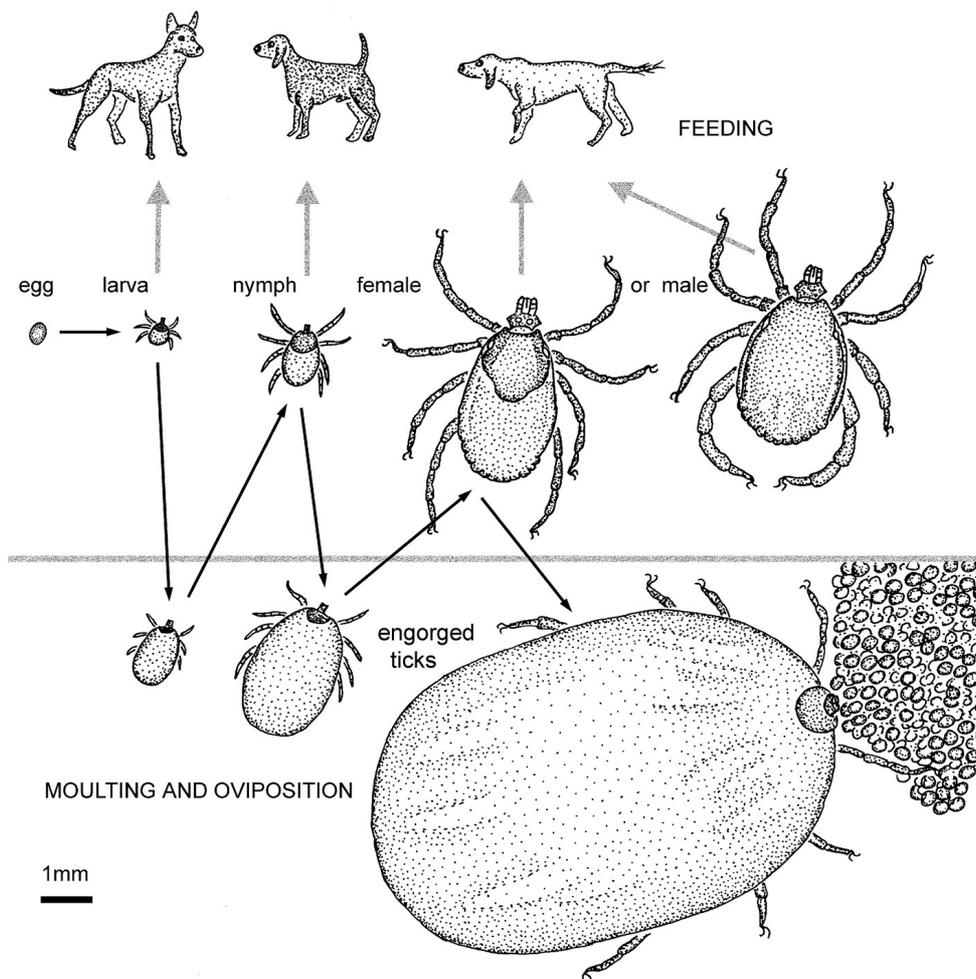


FIGURE 5. Life-cycle of a three-host tick. Example is *Rhipicephalus sanguineus* (brown dog tick) feeding on dogs. NOTE that the different instars and their appearance when engorged are all drawn to the same scale.

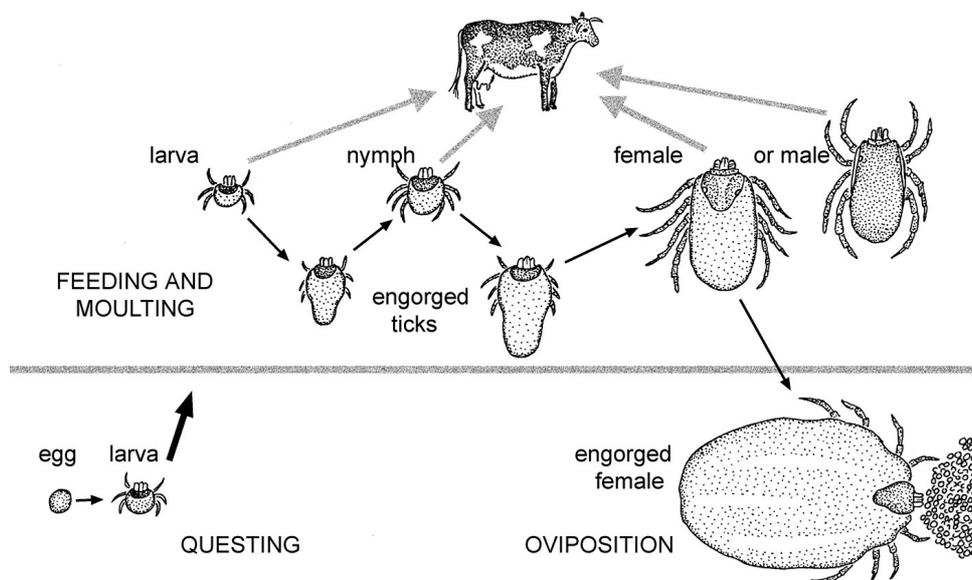


FIGURE 6. Life-cycle of a one-host tick. Example is *Rhipicephalus australis* (Australian cattle tick) feeding on a cow.

Life-cycles of one-host and two-host ticks. Figure 6 shows the sequence of feeding and moulting during the life of individual ticks from a single batch of eggs of a typical one-host species of tick. This is a less common type

of life-cycle than the three-host life-cycle, but it occurs in all species of the subgenus *Boophilus* within the genus *Rhipicephalus* (for example *R. australis*, the Australian cattle tick) and in a few other genera. Eggs are laid in the physical environment. Larvae develop in the eggs until ready to hatch, usually in several weeks. Then the larvae crawl onto vegetation to quest for a host. When they have fed to repletion they remain attached to the host and moulting occurs there. The nymphs then feed on the same individual host and also remain attached to the host. After another moult the adults hatch then feed on the same individual host. The adults will move around the host to find a mate. Thus all three feedings of any one individual tick occur on one individual host. The life-cycle of one-host ticks is usually rapid; for species of the subgenus *Boophilus* it takes three weeks for the feedings on one host and two months for egg laying and hatching of larvae. The two-host life-cycle is similar to the one-host life-cycle but only the larvae and nymphs feed on the same individual host, and the adults will feed on another host of the same species.

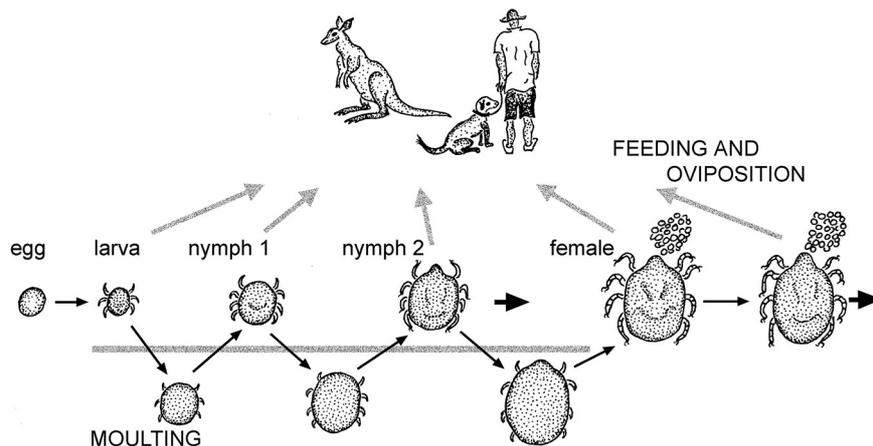


FIGURE 7. Life-cycle of a multi-host argasid tick. Example is *Ornithodoros gurneyi* (kangaroo soft tick) feeding repeatedly at various instars on kangaroos, dogs and humans.

Life-cycles of argasid ticks. Figure 7 shows the sequence of feeding and moulting during the life of individual ticks from a single batch of eggs of a typical argasid (soft) species of ticks. Most argasids have multi-host life-cycles, but *Otobius megnini* (spinose ear tick) has a one-host life-cycle. Larvae either feed rapidly, over a few hours, or feed over several days, then detach from their host and moult. In some species of argasids the larvae do not feed; rather they moult directly to the first nymphal stage without a meal. The first nymphal stage feeds rapidly then moults to a further nymphal stage. Similar feedings follow on different individual hosts and there are often a variable number of nymphal stages. The females feed rapidly on an individual host then lay a small batch of eggs (100 to 500). The females repeatedly feed then lay eggs; up to six feedings and egg layings. Mating occurs off the hosts.

Tick ecology

Habitats and hosts. A tick's habitat is composed of the variety of living and non-living things in the space in which it lives. Ticks are adapted to two contrasting components of their habitat: the physical environment and their host. When ticks are moulting and then questing in the physical habitat they are in danger of drying out and starving. The larvae are most susceptible to drying out and starving because they have a high surface area relative to their small volume. Larvae are also exposed to predators such as rodents, birds, reptiles and ants, and to pathogens such as fungi. These adverse factors limit the type of habitats that a species of tick will be found in, thus knowledge of the typical physical habitats is an aid in the identification of species of ticks. The most important component of the physical environment of a tick is the climate that is defined by temperature and humidity. When the same tick is on the host it is no longer in danger of drying out or starving, but is in danger of being removed by the host's grooming or having its feeding reduced by host-immunity. Most ticks have adaptations in their behaviour and physiology of feeding to reduce these host reactions. Usually these adaptations work best for a certain type of host. The geographic distribution of the potential hosts of a species of tick, however, is often much larger than the distribution of the tick. Ticks feeding on their hosts may also be eaten by domestic chickens and other birds. Most ticks have characteristic species of hosts to which they are adapted. These hosts may be a single species but more commonly are a group of similar species. For example, *Rhipicephalus australis* mainly infests cattle.

How to sample ticks

Collection of ticks from hosts. See Figure 8. Tick specimens are usually collected from their hosts. An effective way to detect adult ticks, especially when they are engorging, is to feel the hair coat of the host with the palm of your hand. Smaller domestic animals in a clinic can be examined in the same way. To find immature ticks or unfed adults, the hair can be parted systematically using forceps as a comb.



FIGURE 8. Sampling of ticks from hosts. (A) Sampling ticks from flank of steer in a squeeze crush. (B) Sampling ticks at udder and groin of a heifer. (C) Sampling ticks on a cast calf. (D) Typical infestation of *Rhipicephalus decoloratus* (the blue tick) adults on belly of a calf. (E) Typical infestation of *Rhipicephalus appendiculatus* (the brown ear tick) on ear of a calf. (F) Infestation of *Amblyomma variegatum* (the tropical bont tick) on dewlap of a steer. [These species of ticks do not occur in Australia but are nonetheless instructive about the biology of the Australian ticks.]

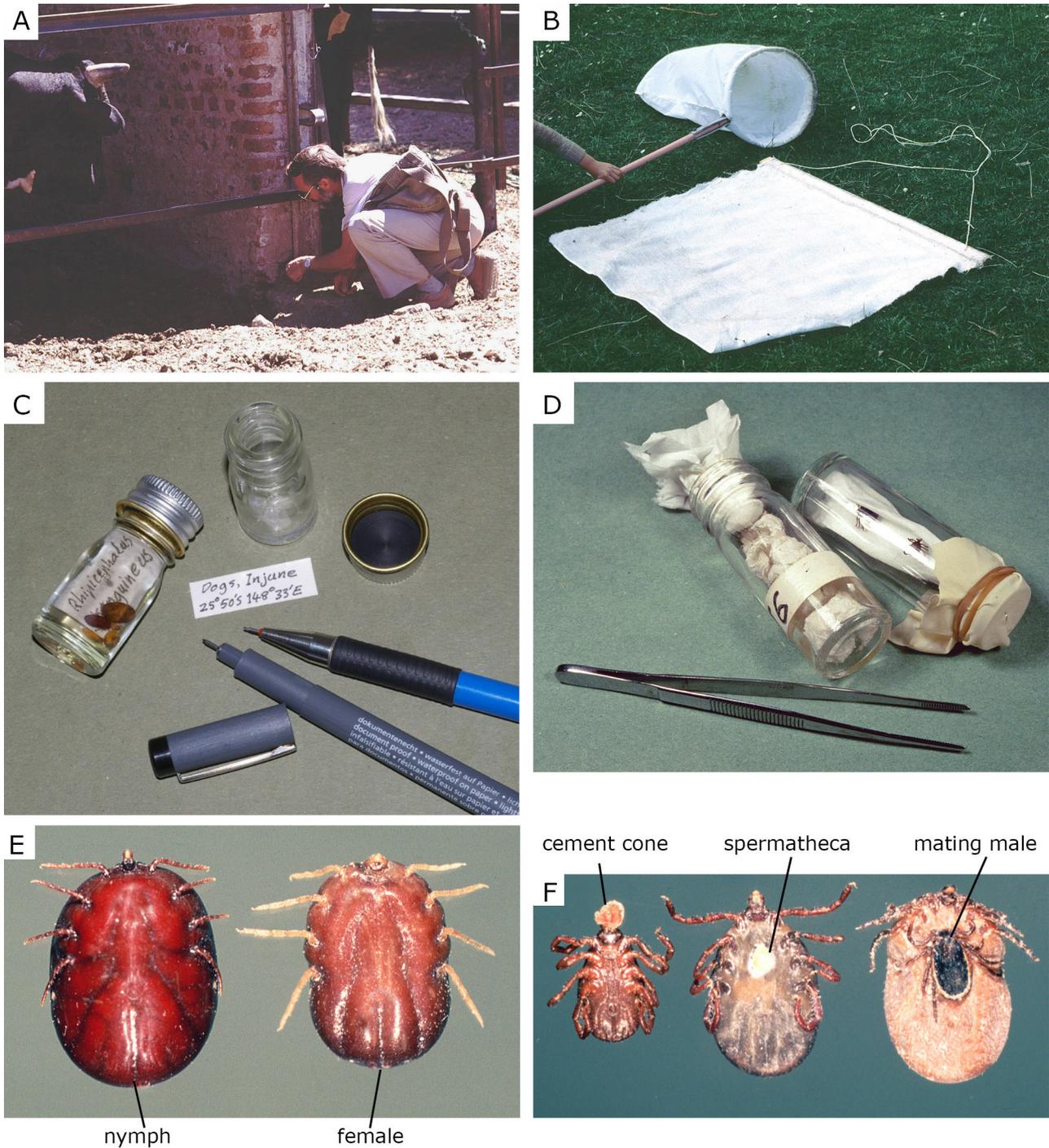


FIGURE 9. Collections of ticks. (A) Collecting *Ornithodoros* and *Hyalomma* adult ticks from the ground and structures of a cattle pen. (B) A sweep net and a blanket drag for collecting ticks from vegetation. (C) Equipment for labelling and preserving ticks. (D) Collecting tubes for live ticks. (E) An engorged nymph of *Amblyomma variegatum variegatum* (the tropical bont tick) and an engorging female *Rhipicephalus microplus* (cattle tick) collected from the same cow at same scale; they can be distinguished by the relatively longer mouthparts of the *Amblyomma*. (F) Features on the ventral surface of ticks confusing for identification. [These species of ticks do not occur in Australia but are nonetheless instructive about the biology of the Australian ticks.]

To remove ticks it is necessary to use strong steel forceps. These should be of medium size with blunt ends and serrated inner surfaces. The forceps are used to grip the tick firmly over its mouthparts only and as closely to the host's skin as possible, then the tick is pulled firmly and directly out. There is no value in twisting the tick as it is removed. Also, commercially available tick removers, such as a plastic spatula with a slit, are effective. It is important to have males in addition to females for identification; males have more diagnostic features. Take care to remove the males, which often re-attach for mating pressed to the ventral side of engorging females, near their genital aperture. If the ticks are required alive they should be placed in strong tubes containing a piece of damp

paper. During collection it is useful to seal the tube with a rubber membrane made from a surgical glove or similar material and held with a rubber band or tape. This should have a small slit cut in it through which the ticks are pushed. For transport to the laboratory use a separate ventilated plug. This can be made of cotton wool or a perforated screw cap. These tubes should be labelled then kept in a sealed plastic bag containing wet cotton wool or paper to maintain high humidity. The ticks should be kept cool over ice but take care not to freeze them. To preserve the ticks at the collection site place them straight into 70 % ethanol (8 parts of 90 % ethanol plus 3 parts of water), or 5 % formalin (5 parts of 40 % formaldehyde solution plus 95 parts water). If the ticks are to be used for any form of analysis of nucleic acids they should be put directly into 100 % laboratory (analytical) grade ethanol and stored in a freezer (if and when possible).

Tick sampling tubes and forceps. See Figure 9. Preserved ticks collected in the field should be placed in 25 to 30 ml capacity glass tubes with thick walls and, ideally, metal screw caps. These are usually known as McCartney or Universal tubes. Their thick glass walls make them more durable than plastic tubes. To label collection tubes in the field the best method is to use lead pencil writing on strong paper or card to make a small label. This is placed inside the tube with the ticks. Labels on the outside of the tubes should only be written on adhesive tape wrapped completely around the tube. Field collection data should include date, site, collector, host species and other information relevant to the study.

Collection of ticks from vegetation and other environments. Some species of ticks can be collected whilst they are unfed and questing on vegetation. If they are sufficiently dense in numbers, adult *Ixodes holocyclus* (paralysis tick) and other ticks can be picked from grass stems. More often it is efficient to use a trap which mimics a host. This consists of a white toweling cloth 1 m square which is dragged slowly across the vegetation for 5 m to 10 m (for approximately 30 seconds of walking; or a longer distance depending on local knowledge). Larvae, nymphs and adults will grip onto the dragging cloth temporarily and can be collected with forceps. The cloth is fitted with a bar at the front and a cord for pulling it. This method works well for larvae and nymphs of questing species but is less efficient for adults and hunting species. Sweep-nets are sometimes used for collecting questing ticks from long vegetation.

Endophilic and domestic ticks can be collected direct from the nests or shelters of their hosts using forceps to probe in cracks and under pieces of dry dung, spiders webs, etc. This is effective for *Argas* ticks. The chitin of ticks tends to be auto-fluorescent in ultraviolet light. This can make ticks easy to see at night if illuminated with a portable ultra-violet lamp. In Australia, most ticks are not a serious risk to humans but in some situations they may be a great risk, especially to people working closely with domestic animals and wildlife. When collecting in the field protect yourself by wearing long trousers over long boots and long sleeves with closures at the wrists.

Preservation and labelling of ticks. Laboratory and museum collections of ticks are stored in liquid. This is usually 70 % ethanol with glycerol for morphological studies (use 90 % ethanol, to 80 ml of this add 15 ml of water and 5 ml of glycerol). The glycerol protects the ticks from drying out when examined. The best tubes for storing ticks are those with thick glass walls and metal screw caps with a rubber washer, of 7 ml capacity and known as Bijou tubes. Plastic containers for storing ticks in a collection may cause problems because some plastics slowly degrade then leak.

Labels for tick collections should be written on card using only India ink (= China ink), which is carbon-based and will not dissolve. A fine draughtsman's pen is necessary for this; modern drawing pens with *insoluble pigment ink* and a fibre-tip of 0.1 mm or 0.2 mm are suitable. The label should include the name of the species (if known), date, collector, species of host and site of collection. The site should be given as both a permanent place name and as latitude and longitude. The use of only small town names or similar changeable features often makes difficulties for later workers. *The universally accepted system is to use global coordinates of latitude and longitude, to at least the nearest minute.* These are read from a map or atlas of the area, or from a global position receiver, eg an iPhone.

Observing ticks

Ticks can be identified to genus using the naked eye or a simple hand lens of x10 magnification. To identify most species, however, a dissecting microscope is needed or at least a higher magnification hand lens. A dissecting microscope is a low power stereoscopic microscope. It must have a range of magnification from x10 to x40, and preferably up to x80. Good lighting is essential; this requires a special microscope lamp. The best type has a cold light source from a bulb of high intensity and is fitted with a flexible light guide. Alternatively, small domestic lamps with flexible stems and strong light emitting diode bulbs are adequate. For preliminary sorting, keep the

ticks in a dish under the preserving liquid. For some features, such as leg colouration, observe the ticks under liquid. For final identification examine ticks dry and cleaned of deposits of glycerol. Use tissue paper to blot the ticks dry. When dry, the ticks often reveal dirt on their surface. Clean them using an ultrasonic cleaner whilst they are immersed in 5 % sodium or potassium hydroxide solution in water. If an ultrasonic cleaner is not available, then get a very fine artist's brush and cut the bristles down to a small stump. Immerse the tick in a solution of detergent and use this stump to clean the tick. If cement adheres to the mouthparts, use fine forceps to pull it off whilst gripping the tick with medium forceps. It is most important to view the tick from different angles to observe features of superficial texture such as punctations and grooves. Use a piece of artist's modelling material such as 'Plasticine' or "Blu-tac" which can be fixed to the bottom of a small dish and moulded into a stand of various angles on which to view the tick. If a hot light-source is used it will dry out the tick and make it brittle. To avoid this, replace the preservative regularly, or have the stand surrounded with water and place a lid over the viewing dish when it is not in use.

The relative sizes of larvae, nymphs and adults when unfed and fed are shown in Figure 6 of the life-cycle of *Rhipicephalus australis*, the Australian cattle tick. It is most important to be familiar with these different sizes. Engorged ticks are difficult to examine but most of the features can be seen if the tick is positioned on a viewing stand. Cement or a spermatheca or a mating male may be found attached to feeding females and should be removed. An engorged *Amblyomma* nymph can be the same size as an engorged *Rhipicephalus australis* adult female.

External structure. Figures 3 shows the main features of the external structure (= morphology) of ixodid ticks. Larvae have three pairs of legs, a scutum covering the anterior dorsal surface and no genital aperture. Nymphs have four pairs of legs, a scutum and no genital aperture. Females have four pairs of legs, a normal scutum and a large genital aperture. Males have four pairs of legs, a conscutum covering most of the dorsal surface and a genital aperture in the same position as the female. Larvae and nymphs can usually be placed in the correct genus by comparison with the mouthparts and coxae of adults. Identification to species of immature ticks is work for an expert but it is helpful for identification of immature ticks to species if they are closely associated with the most abundant species of adult. Coloured patterns occur on some ticks as a pigment (= enamel) in the outer body wall. This colour, however, has limited use for identification of ticks, except in *Amblyomma* and *Dermacentor* species where it may form a distinctive pattern.

Genera and species of ticks

Globally, within the argasid (soft) ticks there are four genera, and within the ixodid (hard) ticks there are 12 extant (living) and three extinct (fossil) genera (Guglielmone *et al.* 2010). Of the extant 16 genera, 11 are illustrated in Figures 10 to 18; including three genera (*Dermacentor*, *Hyalomma* and *Margaropus*) that are not known to occur in Australia but are included here to illustrate some important general features of ticks useful for identification and to expand the readers awareness of other ticks important in animal and human diseases. The other five genera that occur elsewhere (outside of Australia) are *Anomalohimalaya* (3 spp.), *Antricola* (17 spp.), *Cosmiomma* (1 spp.), *Nosomma* (2 spp.) and *Rhipicentor* (2 spp.).

Procedure to identify a tick

Genus. The first stage in identifying ticks is to determine which family and genus they belong to. Then sort the ticks into the main taxonomic groups listed below, according to the morphological features (characters) which are described in the Glossary and in Figures 2, 3 and 10 to 18.

Group comprising *Argas*, *Ornithodoros* and *Otobius*

Size of adults: large (6–7 mm)

Mouthparts: ventral and short.

Other features: scutum or conscutum absent; pulvilli absent.

Group comprising *Amblyomma* and *Bothriocroton*

Size: large (6–7 mm)

Mouthparts: anterior and long.

Other features: anal groove posterior to anus, eyes present in *Amblyomma* species, eyes absent in *Bothriocroton*; pale rings on legs; pale patches on scutum [note that eyes are not present in some *Amblyomma* species from other countries]

The genus *Ixodes*

Size: small to medium (2–5 mm)

Mouthparts: anterior and long in females but short in males.

Other features: anal groove loops anterior to anus, eyes absent; legs usually plain and dark but may vary; males with flat sclerotized plates ventrally.

Group comprising *Haemaphysalis* and *Rhipicephalus*

Size: small to medium (2–5 mm)

Mouthparts: anterior and short.

Other features: anal groove posterior to anus, eyes absent in *Haemaphysalis*, present in *Rhipicephalus*.

When your specimens can provisionally be placed in one of the groups above, then compare the features for the relevant genera illustrated in Figures 10, 11, 12, 14, 16, 17 and 18. If at all possible, examine at least several adult specimens of both sexes. Each sex has some separate and distinctive features useful for identification. Adults are usually easiest to collect and are far easier to identify to species than are larvae or nymphs.

For the argasid genera (*Argas*, *Ornithodoros* and *Otobius*), it is necessary to examine in detail the general shape of the body with or without a suture line between dorsal and ventral surfaces, and the texture of the integument with mammillae or spines. To distinguish the species of *Amblyomma* and *Bothriocroton* of Australia, look for the presence or absence of eyes and the width of the mouthparts relative to the width of the basis capituli. *Ixodes* is the largest genus of ticks with 241 species widely spread in the world and feeding on many species of host; they also have some distinctive features. *Ixodes* is the only genus with a distinct anal groove that extends from the posterior body margin *anteriorly* around the anus, in both sexes. The males are smaller than the females and because they do not feed on vertebrate hosts, the mouthparts of male ticks are much reduced in size (some *Ixodes* species are parasitic on engorging females with which they will also mate eg *I. holocyclus*, the paralysis tick). The males also have a distinctive set of sclerotized plates on their ventral surface. The mouthparts of *Ixodes* females are distinctive with the splayed out first article of the palps protruding beyond the margin of the basis capituli, then the anterior of the palps tapering inwards to give a pointed appearance to the mouthparts. *Margaropus* is a highly distinctive genus that is found only in Africa, which, although small, has extraordinary legs: long and bandy in the females, and with greatly thickened segments in the males. The genus *Rhipicephalus* is most easily distinguished from *Haemaphysalis* by its palps and piercing mouthparts which have a narrower profile than its basis capituli, also by the presence of eyes and of sclerotized plates around the anus of males. *Haemaphysalis* species are smaller than most species of *Rhipicephalus*.

The feature of enamel or ornamentation patterns on ticks can be confusing. This is a superficial pigmentation of the integument of the scutum, conscutum or sometimes parts of the legs, although on the legs it is separate from any general pale versus dark colour of the integument of the segments. Figure 13 shows the typical white enamel on *Dermacentor* ticks (which do not occur in Australia). In Africa, several *Rhipicephalus* species have white enamel, and many *Amblyomma* species have vivid patterns of white, orange and green pigments. Enamel on Australian ticks is more discrete, however (see the small white patch at the posterior of the scutum of female *Amblyomma* and *Bothriocroton* in Figures 11 and 12). *Hyalomma* ticks highlight many of the features used to identify genera, as seen in Figures 3 and 15. The latter figure illustrates *Hyalomma dromedarii*, a serious pest of the dromedary camels of Africa and the Middle East, but unknown on the camel population of Australia.

Species. To identify the species of your specimens proceed to the relevant part of the species descriptions which follow (Figs 19 to 70). For each genus represented here the morphological features used to separate the species have been defined in character matrices (Table 1a–1h). This table has the characters and character-states that are shown on the figures 19 to 70. Each character is sub-divided into the two or more character-states in which the character can occur. For every cell of the table there is either a positive or negative designation. (This strict binary nature of character matrices is ideal for computerisation.) All these characters and their character-states are described in the glossary (Figs 1a–1m). Examine both sexes, both sides, and if possible examine at least several different specimens of each.

Of great value in identification is also the clinical context of host species, geographic distribution, and often any associated disease in their hosts. To distinguish *Ixodes cornuatus* (southern paralysis tick) from *I. holocyclus* (paralysis tick) also consult, but do not rely on, the distribution maps (Figs 49 and 58). To distinguish *Rhipicephalus australis* and *R. sanguineus* remember the common names of these ticks of high host specificity; the Australian cattle tick and brown dog tick, respectively. Beware, however, that sometimes cattle ticks will attach to dogs and brown dog ticks will attach to cattle where both tick and host species are present on a property (farm). Furthermore, the global distribution of *R. sanguineus*, within latitudes approximately 50° north and 40° south, follows closely that of domestic dogs with their human owners whereas in Australia, *R. australis*, the Australian cattle tick is confined to a wide eastern and northern region (see Fig. 67).

Character matrices

TABLE 1A. Character matrix for the family Argasidae genera: *Argas*, *Ornithodoros* and *Otobius*.

Character	State	<i>Arg.</i>	<i>Orn.</i>	<i>Oto.</i>
Eyes	always absent	+		+
	usually absent		-/+	
Margin between dorsal and ventral surfaces as a suture line	absent		+	+
	present	+		
Mammillae and / or ridges on body surface	absent			+
	present	+	+	
Spines covering most of body surface	absent	+	+	
	present			+

NOTES: Eyes occur in some species of *Ornithodoros*; such as *O. savignyi* (not found in Australia);

-/+ means some species are exceptions to the state.

Character-states for *Otobius* refer to the feeding nymph stage only.

TABLE 1B. Character matrix for the *Argas persicus* (poultry tick) and *A. robertsi* (Robert's bird tick).

Character	State	<i>A.per.</i>	<i>A.rob.</i>
Mammillae at lateral suture	large, rectangular	+	
	small, various shapes		+
Ridges on integument	sparse		+
	common	+	
Mammillae on integument	sparse	+	
	common		+

TABLE 1C. Character matrix for *Ornithodoros gurneyi* (kangaroo soft tick) and *O. capensis* (seabird soft tick).

Character	State	<i>O.cap.</i>	<i>O.gur.</i>
Tarsal dorsal humps	absent	+	
	present		+
Anterior projection of body	absent		+
	present	+	
Discs on main body	absent		+
	present	+	

..... continued on the next page

TABLE 1C (continued)

Character	State	<i>O.cap.</i>	<i>O.gur.</i>
Ridged areas on main body	absent	+	
	present		+
Postpalpal setae	absent		+
	present	+	
Camerostome cheeks each side	single flap	+	
	multiple flaps		+
Transverse postanal groove	indistinct and single	+	
	distinct and paired		+
Setae on ventral surface	sparse	+	
	dense		+

TABLE 1D. Character matrix for genera of the family Ixodidae: *Amblyomma*, *Bothriocroton*, *Haemaphysalis*, *Ixodes* and *Rhipicephalus*.

Character	State	<i>Amb.</i>	<i>Bot.</i>	<i>Hae.</i>	<i>Ixo.</i>	<i>Rhi.</i>
Body profile from above (unfed ticks)	narrow oval			+	+	+
	broad oval to circular	+	+			
Eyes	absent		+	+	+	
	present	+				+
Festoons at posterior body margin (unfed ticks)	absent				+	
	present	+	+	+		-/+
Sclerotized plates aligned with anus (males only)	absent	+	+	+		
	present				+	+
Anal groove position	anterior to anus				+	
	posterior to anus	+	+	+		+
Mouthparts length relative to basis capituli length	shorter or same length			+		+
	longer	+	+		+	
Mouthparts width relative to basis capituli	narrower or equal	+	+			+
	wider			+	+	
Enamel patterns on scutum or conscutum	absent			+	+	+
	present	+	+			
Leg colour pattern as pale rings	absent			+	+	+
	present	+	+			

NOTES: Festoons occur in most species of *Rhipicephalus* but not in *R. australis* (Australian cattle tick).
-/+ means some species are exceptions to the state.

TABLE 1E. Character matrix for *Bothriocroton auruginans* (wombat tick) and *B. hydrosauri* (southern reptile tick).

Character	State	<i>B.aur.</i>	<i>B.hyd.</i>
Porose area position	narrowly separated and touching margin of basis capituli	+	
	widely separated and above margin of basis capituli		+
Cervical grooves	shallow anteriorly	+	
	deep anteriorly		+

..... continued on the next page

TABLE 1E. (continued)

Character	State	<i>B.aur.</i>	<i>B.hyd.</i>
Enamel on scutum of female	none	+	
	small patch near posterior margin		+
Setae on alloscutum	thin, colourless and sparse	+	
	broad, white and numerous		+
Tarsal dorsal humps	small		+
	large	+	
Coxae 1 anterior projection	small and not visible dorsally		+
	large and visible dorsally	+	
Columns of teeth on hypostome of female	2 + 2	+	
	4 + 4		+
Columns of teeth on hypostome of male	2 + 2	+	
	3 + 3		+
Marginal groove of male	formed by punctations, continuing to first festoons	+	
	formed by a groove, continuous around all festoons		+
Festoon grooves of male	narrow	+	
	broad		+
Ventral plaques on males	absent	+	
	present		+
Punctations on conscutum	indistinct and sparse	+	
	distinct and common		+

TABLE 1F. Character matrix for *Haemaphysalis bancrofti* (wallaby tick) and *H. longicornis* (bush tick).

Character	State	<i>H.ban.</i>	<i>H.lon.</i>
Palp article 2 lateral projection	small		+
	large	+	
Palp article 2 external margin	slightly concave		+
	distinctly concave	+	
Palp article 3 dorsal posterior spur (females)	absent	+	
	present		+
Palp article 3 dorsal posterior spur (males)	small	+	
	large		+
Scutum posterior margin (females)	smoothly rounded	+	
	slightly angular		+
Scutum texture	smooth		+
	corrugated	+	
Cervical grooves	shallow anteriorly	+	
	deep anteriorly		+
Scutum punctations density	sparse	+	
	moderate		+

..... continued on the next page

TABLE 1F (continued)

Character	State	<i>H.ban.</i>	<i>H.lon.</i>
Festoons of female enclosed by lateral groove	1		+
	2	+	
Festoons of male enclosed by lateral groove	0		+
	1	+	
Coxa I external spur	bluntly pointed	+	
	sharply pointed		+
Columns of teeth on hypostome	4 + 4	+	
	5 + 5		+
Spiracle plate shape in males	oval	+	
	like a comma		+

TABLE 1G. Character matrix for *Ixodes holocyclus* (paralysis tick), *I. cornuatus* (southern paralysis tick), *I. tasmani* (common marsupial tick) and *I. hirsti* (Hirst's marsupial tick).

Character	State	<i>Ix.cor.</i>	<i>Ix.hir.</i>	<i>Ix.hol.</i>	<i>Ix.tas.</i>
Palp article 1	bulbous, distant from base of cheliceral sheath	+	+	+	
	widely angular, covering base of cheliceral sheath				+
Palpal shape	narrowly pointed	+	+	+	
	widely angular				+
Hypostome shape	narrowing anteriorly	+	+	+	
	widening anteriorly				+
Basis capituli dorsal posterior margin	straight				+
	undulating	+	+	+	
Cornua on dorsal basis capituli	absent		+		+
	present	+		+	
Scutum proportion	longer than wide	+	+		
	wider than long			+	+
Body profile (males only)	broadly oval	+		+	
	narrowly oval		+		+
Punctations on scutum/conscutum; size	small	+	+	+	
	large				+
Punctations on scutum/conscutum; distribution	sparse	+	+		
	dense			+	+
Scapular grooves	absent				+
	present	+	+	+	
Cervical groove length	short		+	+	
	long	+			+
Lateral grooves on conscutum	absent				+
	present	+	+	+	
Scutal corrugations at posterior	absent		+		+
	present	+		+	

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TABLE 1G (continued)

Character	State	<i>Ix.cor.</i>	<i>Ix.hir.</i>	<i>Ix.hol.</i>	<i>Ix.tas.</i>
Auriculae on ventral basis capituli	absent				+
	present	+	+	+	
Genital aperture position	level with coxae 2 or 3				+
	level with coxae 4	+	+	+	
Sternal plate	absent	+		+	+
	present		+		
Anal groove shape	open at posterior				+
	joined (or closed) at posterior	+	+	+	
Coxa 1 external spurs	absent				+
	present	+	+	+	
Coxa 1 internal spurs (males only)	absent	+		+	+
	present		+		
Coxae type	without syncoxae	+	+	+	
	with syncoxae				+
Trochanter spurs (males only)	indistinct		+	+	+
	distinct	+			
Coxal ridges	absent	+		+	+
	present		+		
Spiracular plate shape	circular	+	+	+	
	oval				+
Posterior grooves (median and lateral) of unfed females	absent	+	+	+	
	present				+
Leg colour pattern	none	+	+		+
	legs 1 and 4 darker than legs 2 and 3			+	
Tarsi terminal profile	gradually stepped			+	
	steeply stepped	+	+		+

TABLE 1H. Character matrix for *Rhipicephalus sanguineus* (brown dog tick) and *R. australis* (Australian cattle tick).

Character	State	<i>R.san.</i>	<i>R.aus</i>
Columns of teeth on hypostome	3 + 3	+	
	4 + 4		+
Eye size and profile	small and flat		+
	large and slightly convex	+	
Punctations on conscutum or scutum	not visible		+
	conspicuous	+	
Scutum posterior margin	single concavity each side		+
	double concavity each side	+	
Cervical fields of female	shallow and indistinct		+
	deep and distinct	+	

..... continued on the next page

TABLE 1H (continued)

Character	State	<i>R.san.</i>	<i>R.aus</i>
Spiracle plate shape	circular		+
	like a comma	+	
Festoons	absent		+
	present	+	
Coxa 1 anterior spur of male	small, not visible dorsally	+	
	large, visible dorsally		+
Setae on scutum and alloscutum	short and sparse	+	
	long and numerous		+
Lateral grooves	absent		+
	present	+	
Anal groove	indistinct		+
	distinct	+	
Adanal plates shape	broadly curved at posterior	+	
	narrowly angular at posterior		+
Caudal appendage of unfed male	absent	+	
	present		+
Tarsal terminal spurs	absent	+	
	present		+

Further reading:

(i) Anonymous (1984) Ticks and Tick-borne Disease Control. Vols 1 & 2, Food and Agriculture Organisation, Rome; (ii) Bowman & Nuttall (1998), Ticks: biology, disease and control. Cambridge University Press, UK; (iii) Coetzer & Tustin (2004), Infectious Diseases of Livestock, with special reference to Southern Africa. Oxford University Press, Oxford, UK; (iv) Lefèvre *et al.* (2010), Infectious and Parasitic Diseases of Livestock, volumes 1 & 2, CABI, Wallingford, UK; (v) Sonenshine & Roe (2014a, b), Biology of Ticks, volumes 1 & 2, Oxford University Press, New York, USA; and (vi) Walker *et al.* (2003), Ticks of Domestic Animals in Africa: a Guide to Identification of Species. Bioscience Reports, Edinburgh, UK.

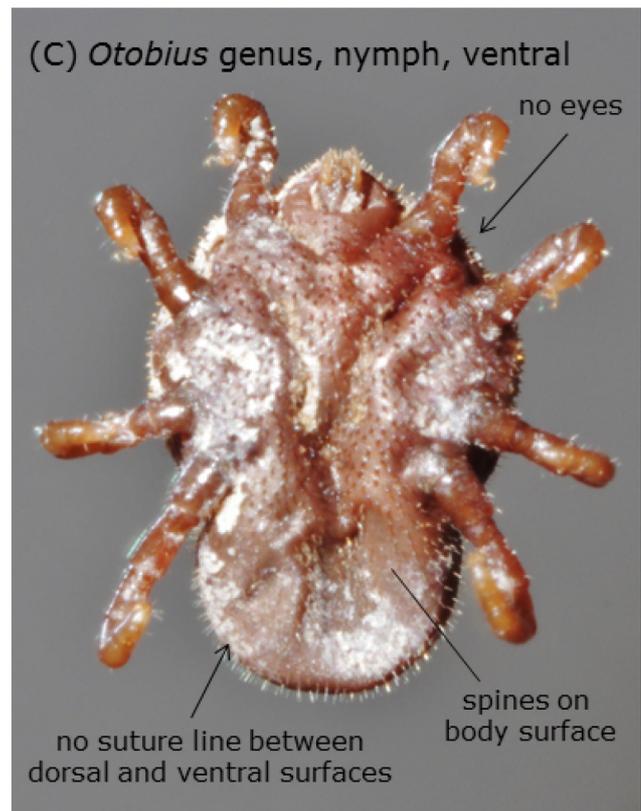
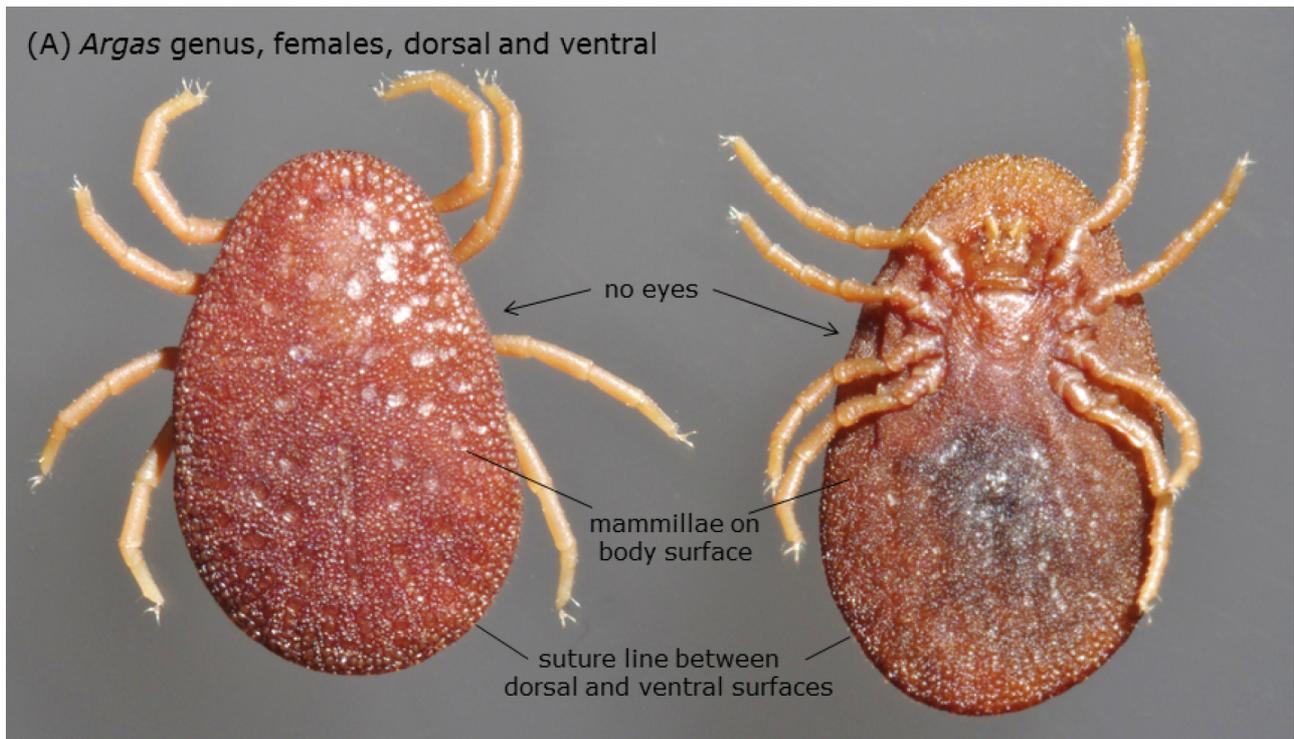


FIGURE 10. Morphological features of genera of the family Argasidae (A) *Argas*, (B) *Ornithodoros* and (C) *Otobius*. Examples are *A. persicus* (poultry tick), *O. capensis* (seabird soft tick) and *O. megnini* (spinose ear tick).

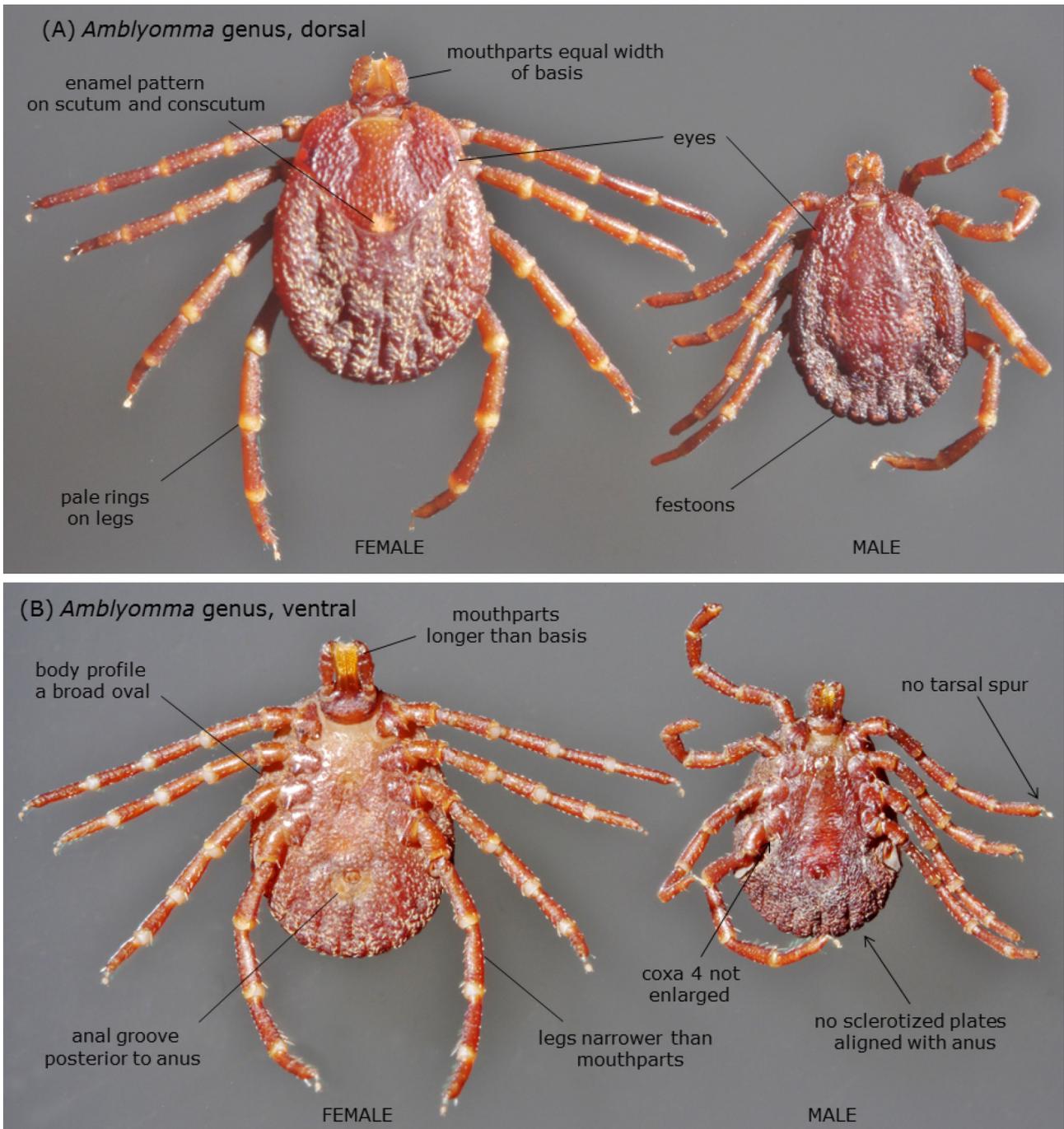


FIGURE 11. Morphological features of the genus *Amblyomma*, (A) dorsal female and male, (B) ventral female and male. Example is *A. triguttatum* (ornate kangaroo tick).

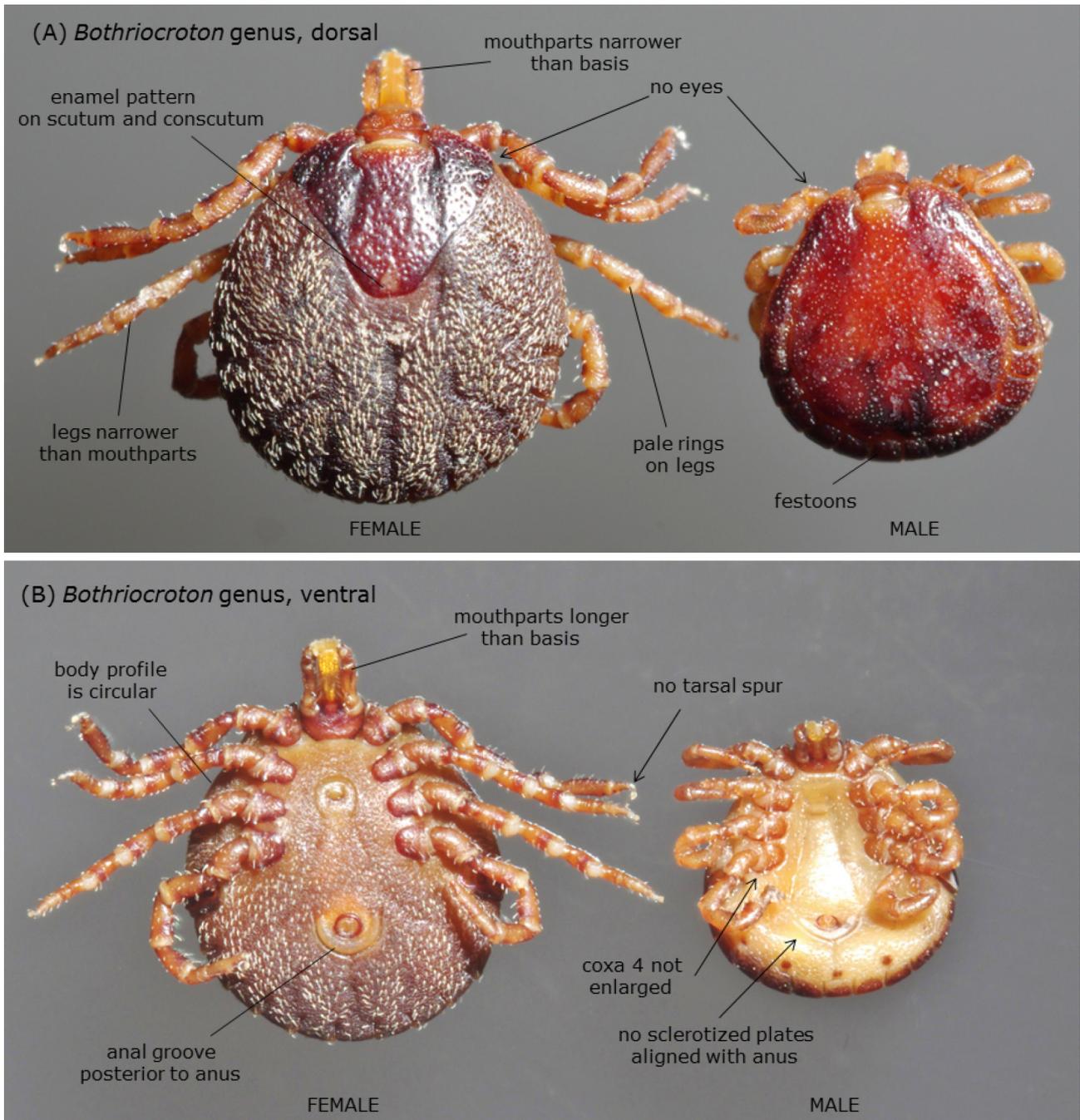


FIGURE 12. Morphological features of the genus *Bothriocroton*, (A) dorsal female and male, (B) ventral female and male. Example is *B. hydrosauri* (southern reptile tick).

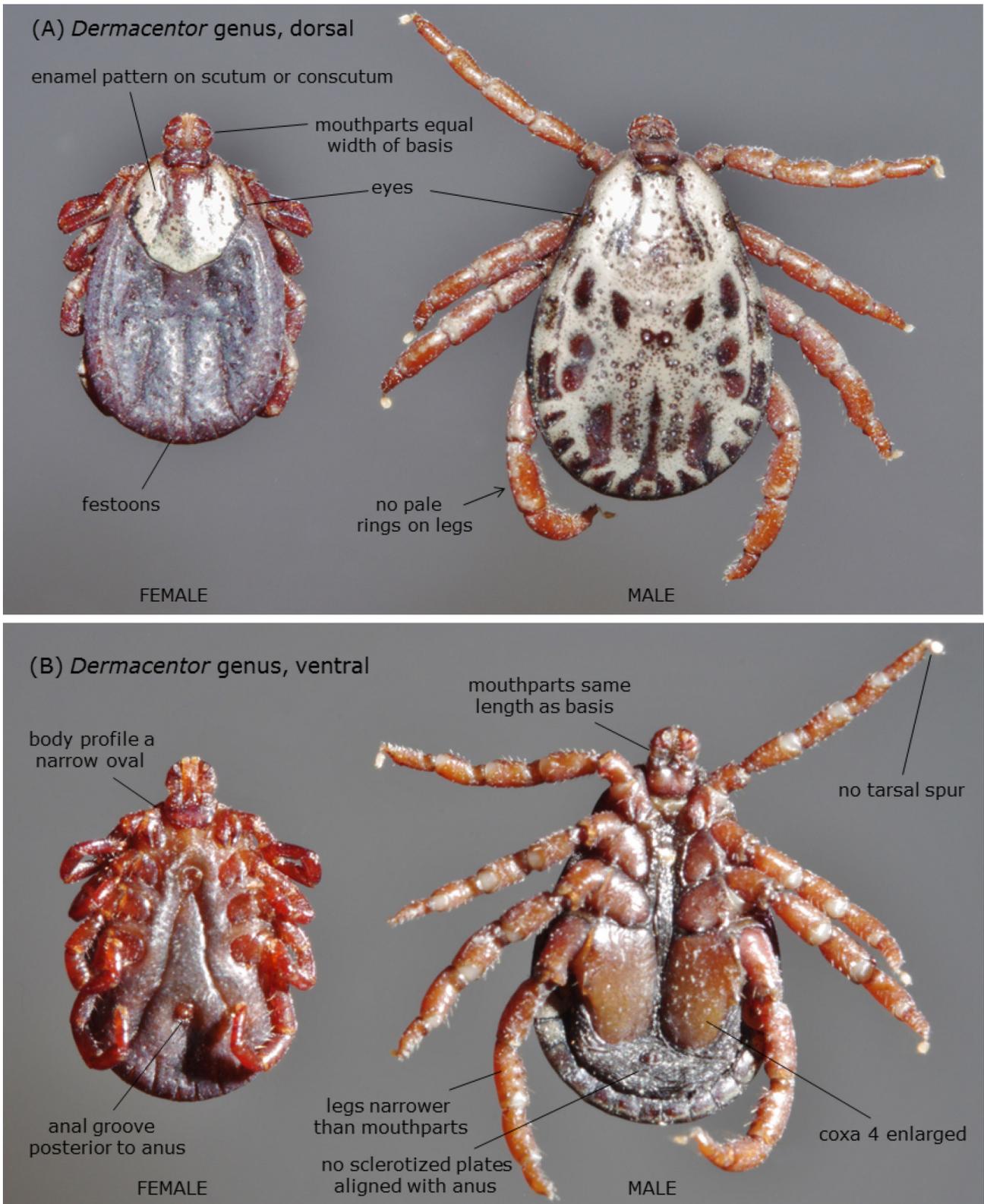


FIGURE 13. Morphological features of the genus *Dermacentor*, (A) dorsal female and male, (B) ventral female and male. Example is *D. andersoni* (rocky mountain wood tick).

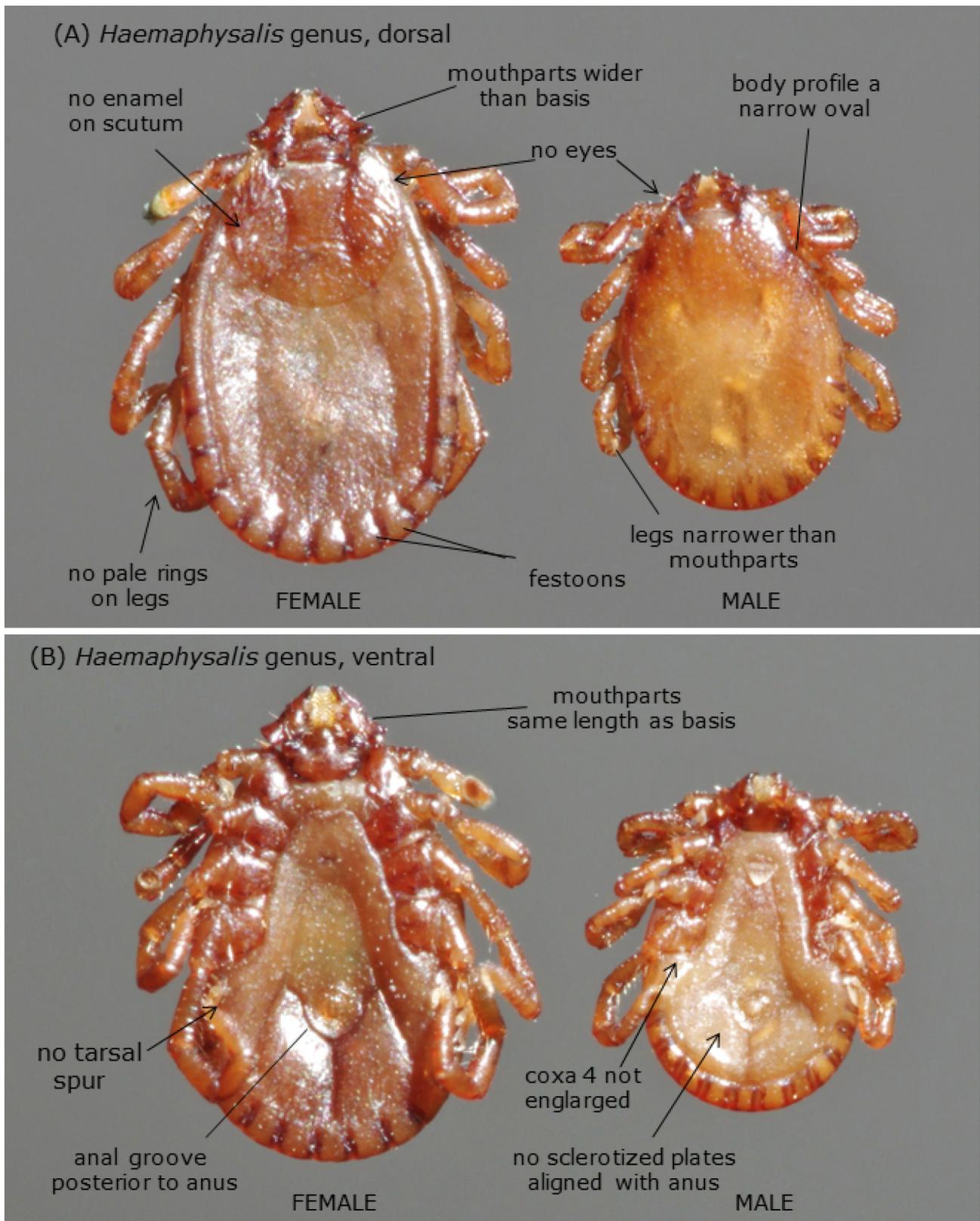


FIGURE 14. Morphological features of the genus *Haemaphysalis*, (A) dorsal female and male, (B) ventral female and male. Example is *H. bancrofti* (wallaby tick).

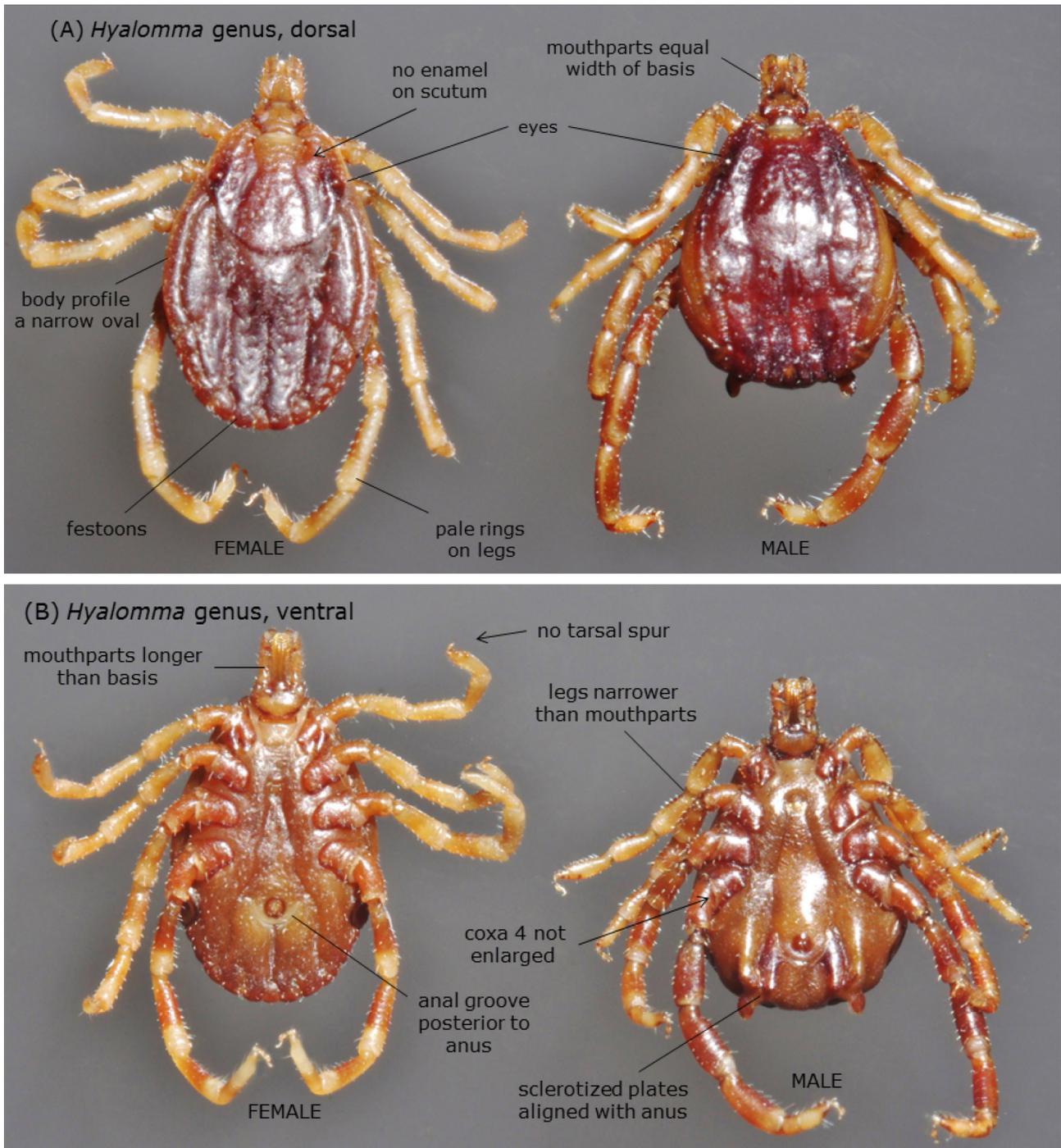


FIGURE 15. Morphological features of the genus *Hyalomma*, (A) dorsal female and male, (B) ventral female and male. Example is *H. dromedarii* (the camel *Hyaloma*).

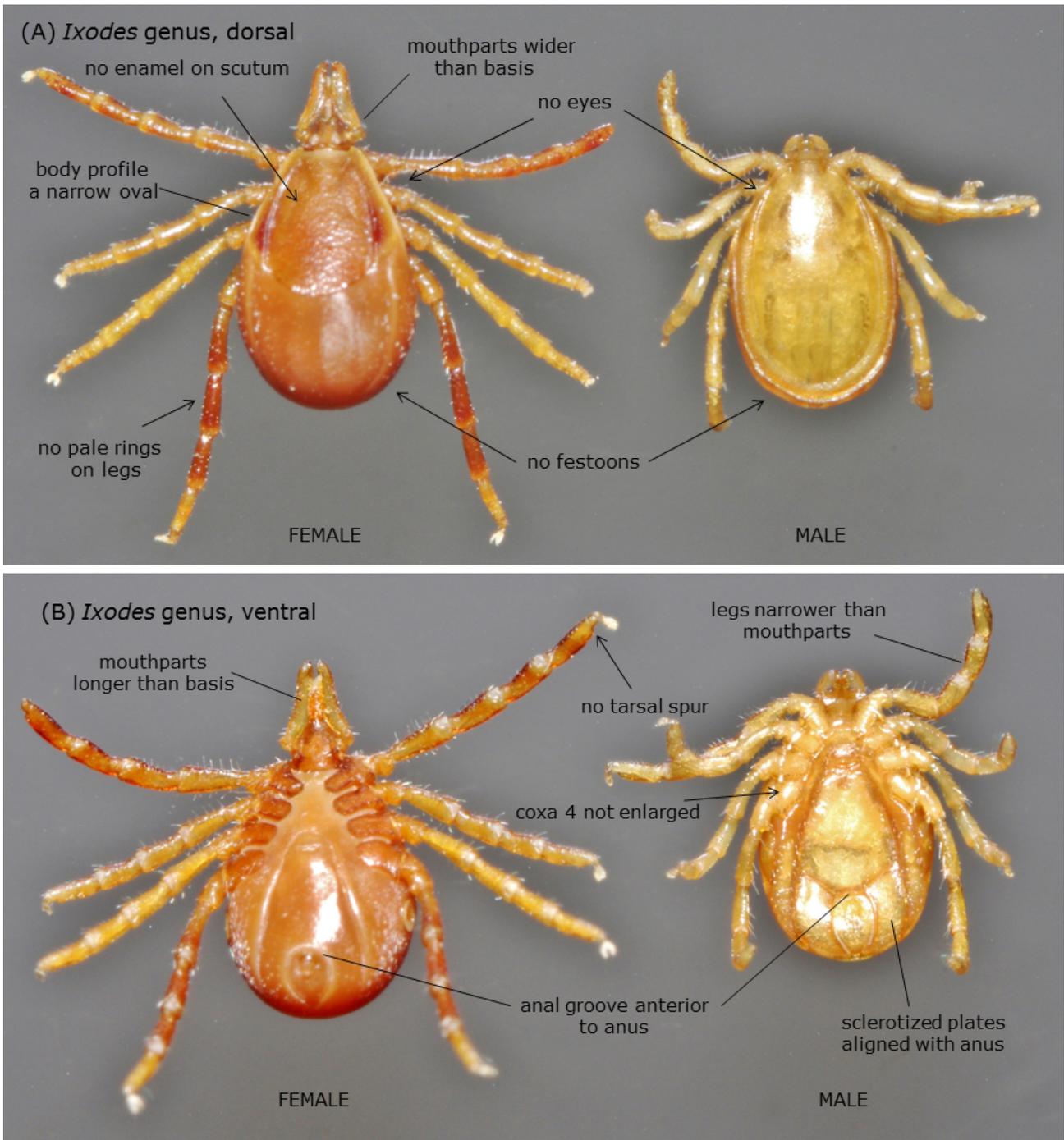


FIGURE 16. Morphological features of genus *Ixodes*, (A) dorsal female and male, (B) ventral female and male. Example is *I. holocyclus* (paralysis tick).

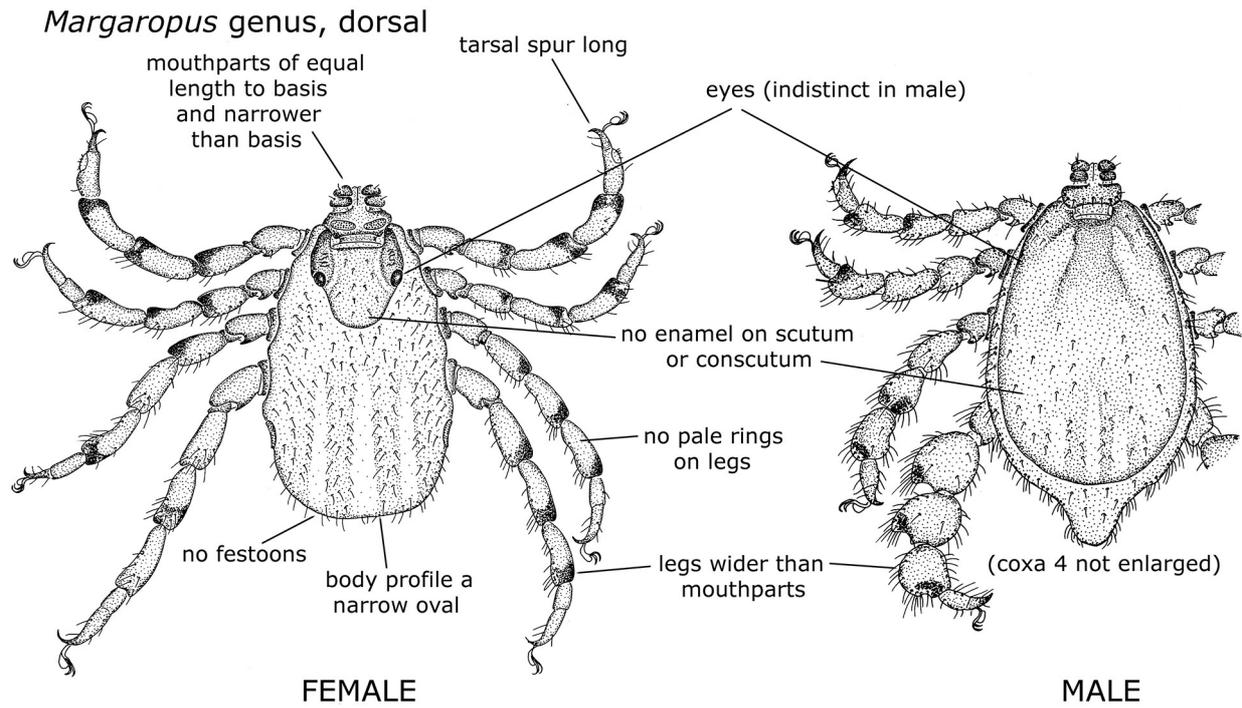


FIGURE 17. Morphological features of the genus *Margaropus*, dorsal views of female and male. Example is *M. winthemi* (the winter horse tick).

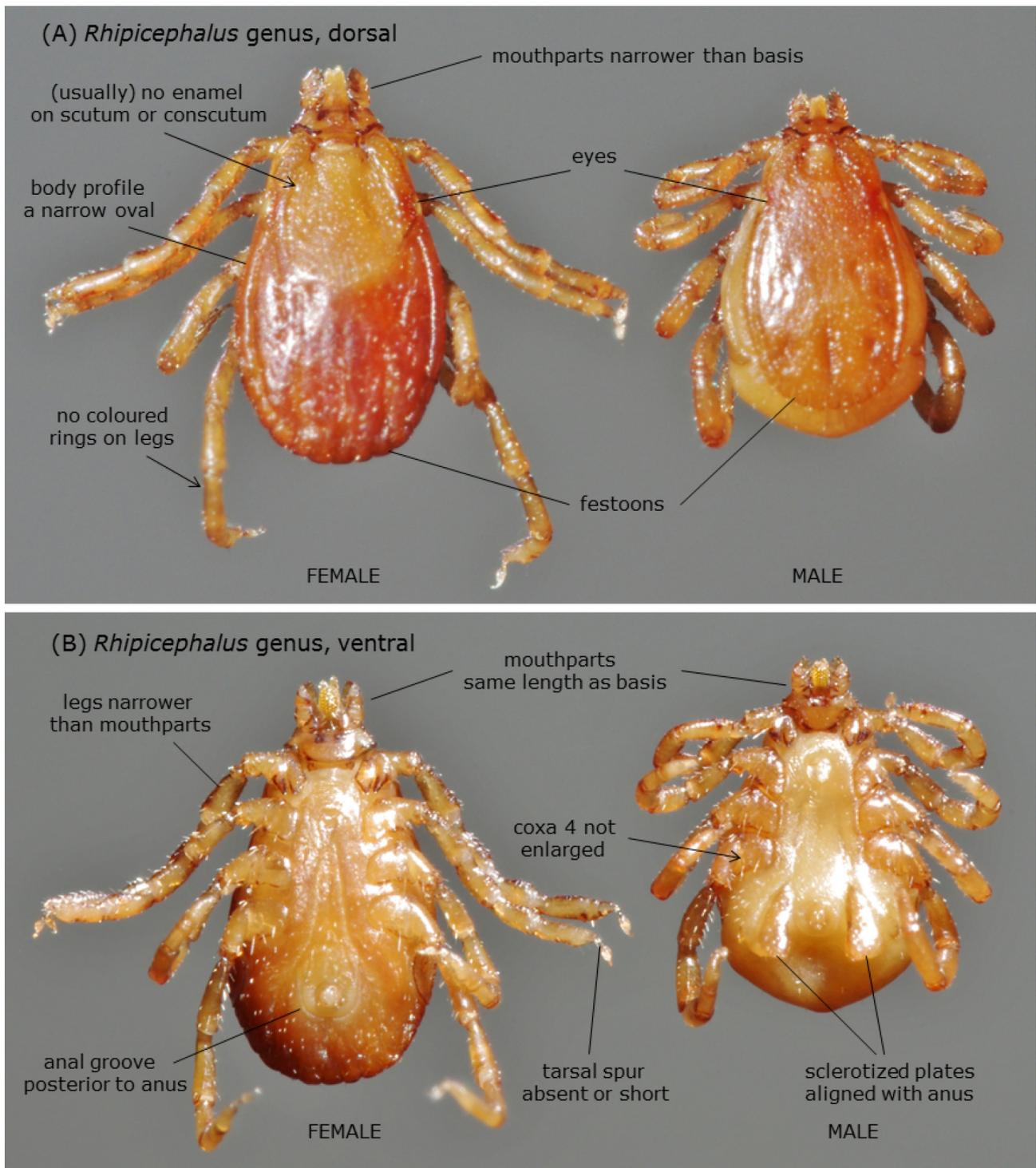
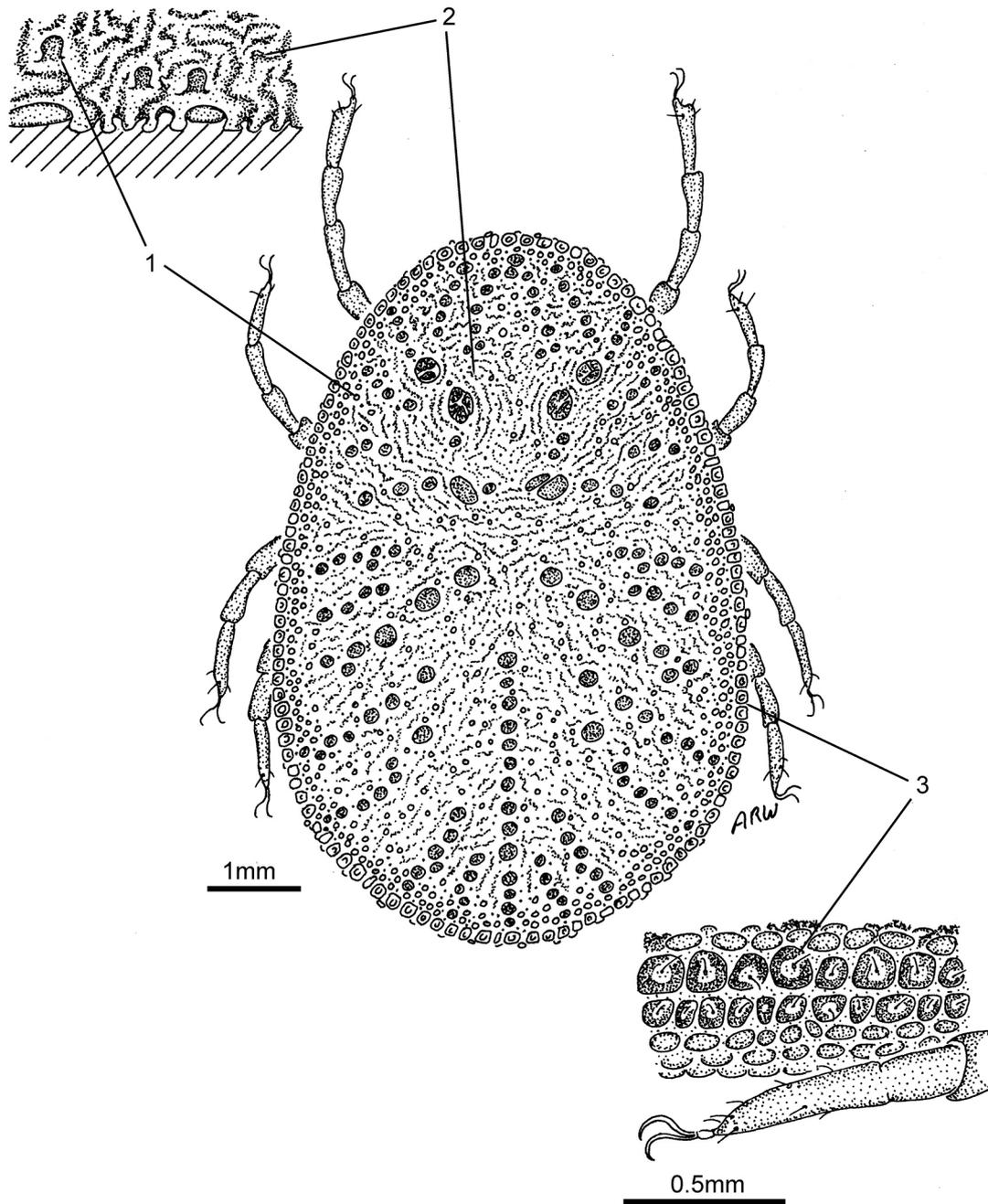


FIGURE 18. Morphological features of the genus *Rhipicephalus*, (A) dorsal female and male, (B) ventral female and male. Example is *R. sanguineus* (brown dog tick).

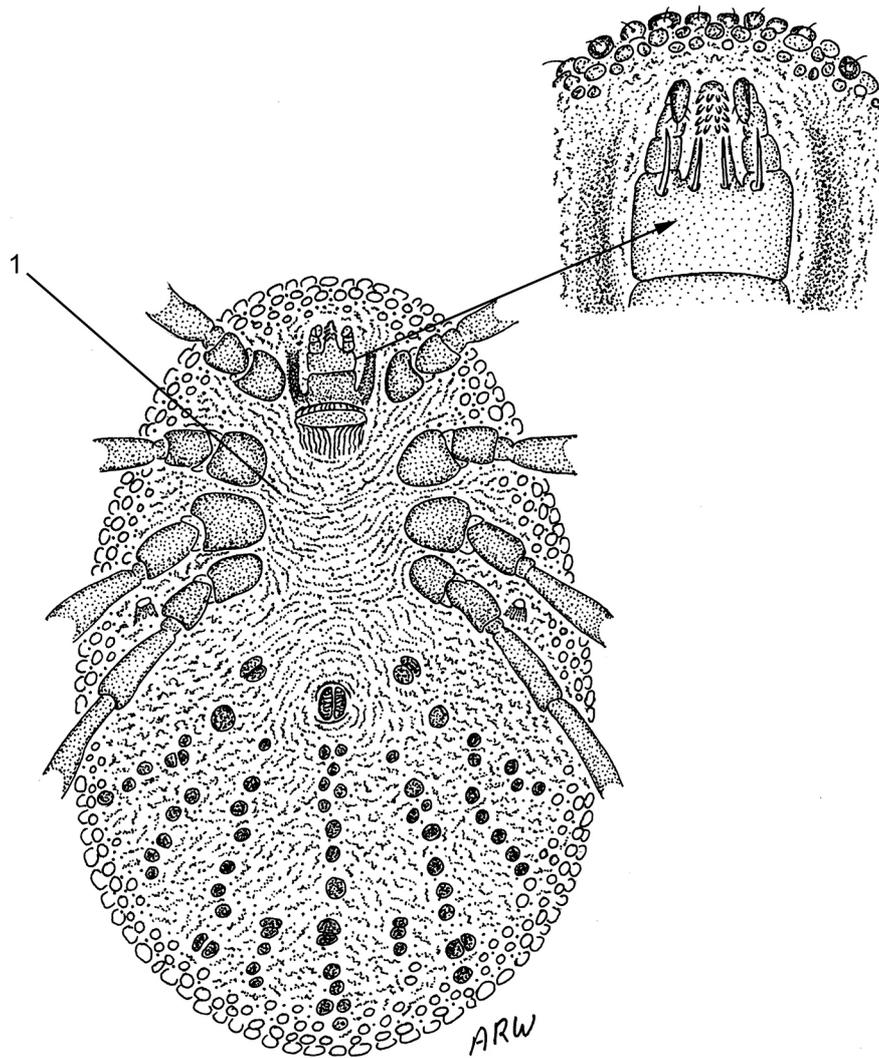
Species accounts

Argas persicus (Oken, 1818) (poultry tick)



- 1 Mammillae on integument are sparse.
- 2 Ridges on integument are common.
- 3 Mammillae at lateral suture are large.

FIGURE 19. *Argas persicus* (poultry tick), female, dorsal.



1 Ridges on integument are common.

FIGURE 20. *Argas persicus* (poultry tick), female, ventral.

General

In Australia, *Argas persicus* is known as the fowl tick or poultry tick; elsewhere it is also known as the fowl tampan (Walker *et al.* 2003) and the Persian poultry *Argas* (Kohls *et al.* 1970). *Argas persicus* is one of three species in the subgenus *Argas* (*Persicargas*) known from Australia: the other two species are *A. (P.) robertsi* (see the next species account) and *A. (P.) nullaborensis* which is known only from the nest of a bird, probably the little crow, *Corvus bennetti* (Hoogstraal & Kaiser 1973).

Differential diagnosis

See Figs 10A, 19 and 20. *Argas* species are, in general, difficult to differentiate. Fortunately only two species of *Argas* are known from domestic animals in Australia, *A. persicus* and *A. robertsi*. *Argas* species grow to become large ticks. *Argas persicus* adults are typically 7.0 mm long by 4.5 mm wide, whereas *A. robertsi* adults are typically 6.0 mm long by 3.5 mm wide. The patterns on the integument are used to differentiate these two species. These patterns are most conspicuous dorsally, and it is necessary to distinguish between mammillae and discs (see Mammillae in the Glossary). *Argas persicus* has sparse mammillae but ridges are common; in addition the

mammillae in *A. persicus* at the lateral suture are large. *Argas robertsi* has common mammillae but the ridges are sparse; in addition the mammillae in *A. robertsi* at the lateral suture are small. Both *A. persicus* and *A. robertsi* have mouthparts characterised by pairs of post-hypostomal setae and post-palpal setae. Identification of male *Argas* species is by the same character-states as for the females. Males tend to be slightly darker and smaller and the genital pore is half the width of that of the female.

Hosts

The only known native (wild) host of *Argas persicus* is the rook, *Corvus frugilegus*, in Central Asia. Central Asia extends from the Caspian Sea in the west, to China in the east, and from Afghanistan in the south to Russia in the north i.e. the five "Stan" nations, Kazakhstan, Kyrgyzstan, Tajikistan, Turkmenistan & Uzbekistan. Recently, however, nymphs and adult *A. persicus* were found under the bark of river red gum trees, *Eucalyptus camaldulensis*, at two sites in Sicily, Italy; poultry were not nearby in either case (Pantaleoni *et al.* 2010). The host of these ticks was not obvious but was apparently a wild bird. Pantaleoni *et al.* (2010) reminds us to look for *Argas* ticks under the bark of trees like river red gums. Pantaleoni *et al.* (2010) concluded that the natural geographic distribution of *A. persicus* includes Italy, northwestern Mediterranean and thus the natural distribution of this tick might be referred to as Central Asia plus northwestern Mediterranean. *Argas persicus* hides under the bark of trees and in other places and thus could easily go undetected in other parts of the Mediterranean (Pantaleoni *et al.* 2010). The main host of *A. persicus* in Australia is the domestic fowl, but *A. persicus* may also infest domestic turkeys and ducks (and we presume geese), and domestic pigeons and canaries (and we presume other cage birds), in Australia (Seddon 1968).

Host records from Roberts (1970): *Serinus canaria* (house canary, introduced), *Gallus gallus* (domestic fowl, introduced), *Anas platyrhynchos* (domestic duck, introduced), *Meleagris gallopavo* (domestic turkey, introduced) and *Columbia livia* (domestic pigeon, introduced).

Host records after Roberts: *Gallus gallus* (domestic fowl, introduced) (Petney *et al.* 2004).

Life-cycle and seasonality

The life-cycle of *A. persicus* is typical of argasid ticks (Walker *et al.* 2003). The females lay batches of 20 to 100 eggs after each blood meal. The eggs hatch in approximately three weeks. The larvae attach and feed on a host for 5 to 10 days, usually under the wings. They detach then moult in cracks and crevices in the poultry house. The nymphs will feed for 5 minutes to a few hours and then moult. There can be four nymphal stages, each requiring a blood-meal before moulting to the next stage. Moulting occurs in cracks and crevices and the moult to the adult can occur from the second nymphal stage onwards. The final nymphal stage moults to the adults; these also feed only for a short while and, like the nymphs, usually feed at night when the birds are roosting. The adults feed about once a month. The females produce a batch of eggs after each blood-meal and they may produce six or seven batches during their lifetime. The larvae can survive for two months or more, the nymphs for one year and the adults for up to three years without a blood-meal. In climates with a winter season, the larvae and first nymphal stage are most active in early summer, nymphal stages two to four in mid-summer, adults late summer and autumn. Ticks over-winter in the adult stage or as eggs.

Disease

Argas persicus is known to transmit to domestic poultry in Australia the bacterium *Aegyptianella pullorum* and the bacterium (spirochaete) *Borrelia anserina* which causes avian spirochaetosis (Roberts 1952, Seddon 1968). Very large populations of ticks can build up rapidly in untreated poultry houses and severe anaemia can develop in poultry. Egg production may be reduced or cease (Roberts 1952). Petney *et al.* (2004) reviewed the pathogenic bacteria and viruses that have been associated with *A. persicus*. Transmission of *B. anserina* by *A. persicus* is transovarial, at least in the laboratory (Gothe *et al.* 1981). *Argas persicus* has also been implicated in the transmission to poultry of *Mycobacterium avian* and *Pasteurella avicida*, and might be involved in the epidemiology of West Nile virus and *Salmonella gallinarum/pullorum*, which is excreted in the faeces of *A. persicus*.

Habitat and geographic distribution

Worldwide, *A. persicus* is found in areas with climates from desert to Mediterranean temperate and to rain forest.

Argas persicus apparently evolved in Central Asia (Palearctic Zoogeographical Region) and then spread to other parts of the world, presumably with domestic poultry. In Australia, *A. persicus* is a domestic and endophilic tick that lives in the fabric of poultry houses and bird nests and other bird-roosting sites. *Argas persicus* is known from all states in Australia except Tasmania (Roberts 1970). Records of *A. persicus* in Tasmania would be noteworthy. *Argas persicus* was first recorded in Australia in 1887. Seddon (1968) has a set of hypotheses for the spread of *A. persicus* in Australia but strong evidence for these hypotheses is not obvious to us.

Genes and genomes

Parts of the following genes are available to the public in GenBank: (i) small subunit mitochondrial rRNA (16S) (ca. 450 bp, Black & Piesman 1994; Petney *et al.* 2004; Pantaleoni *et al.* 2010); (ii) NADH dehydrogenase subunit 5 (519 bp, Chitimia *et al.* 2010); (iii) cytochrome c oxidase subunit I (COX1) (783 bp Chitimia *et al.* 2010); and (iv) small subunit mitochondrial rRNA (12S) (401 bp, Norris *et al.* (1999); and (v) small subunit nuclear rRNA (18S) (1787 bp, Black *et al.* 1997).

Other information

Argas persicus is one of 16 species in the subgenus *Argas* (*Persicargas*) worldwide: comprising the 15 species referred to by Hoogstraal (1985) plus *A. (P.) keiransi* (Estrada-Peña *et al.* 2003). Fifteen of these 16 species parasitise birds that have arboreal nests of sticks and thin branches (Hoogstraal 1985; and Barker unpublished information). In fowl-houses, *A. persicus* and *A. robertsi* shelter in the wood crevices of the building, which are similar to the crevices in the arboreal stick-nests of their native hosts (Hoogstraal, 1985). Six species of the subgenus *Persicargas* have adapted to domestic fowl from life in the arboreal nests of their wild hosts: *A. (P.) persicus*, *A. (P.) robertsi*, *A. (P.) miniatus*, *A. (P.) radiatus*, *A. (P.) arboreus* and *A. (P.) walkerae* (Hoogstraal 1985).

Petney *et al.* (2004) showed that *A. persicus sensu stricto* does occur in southern Australia and is the only species of *Argas* in southern Australia recorded, so far, from poultry. Burger *et al.* (2014a, Table 1) compared the alternative classification schemes for the family Argasidae (soft ticks) and inferred the phylogenetic position of *A. persicus* within the family Argasidae. Seddon (1968) has information on the control of *A. persicus* in fowl-houses. Moorhouse (1975) provided a comprehensive laboratory study of the feeding of larval *A. persicus* on fowls in Brisbane, Australia.

***Argas robertsi* Hoogstraal, Kaiser and Kohls, 1968 (Robert's bird tick)**

General

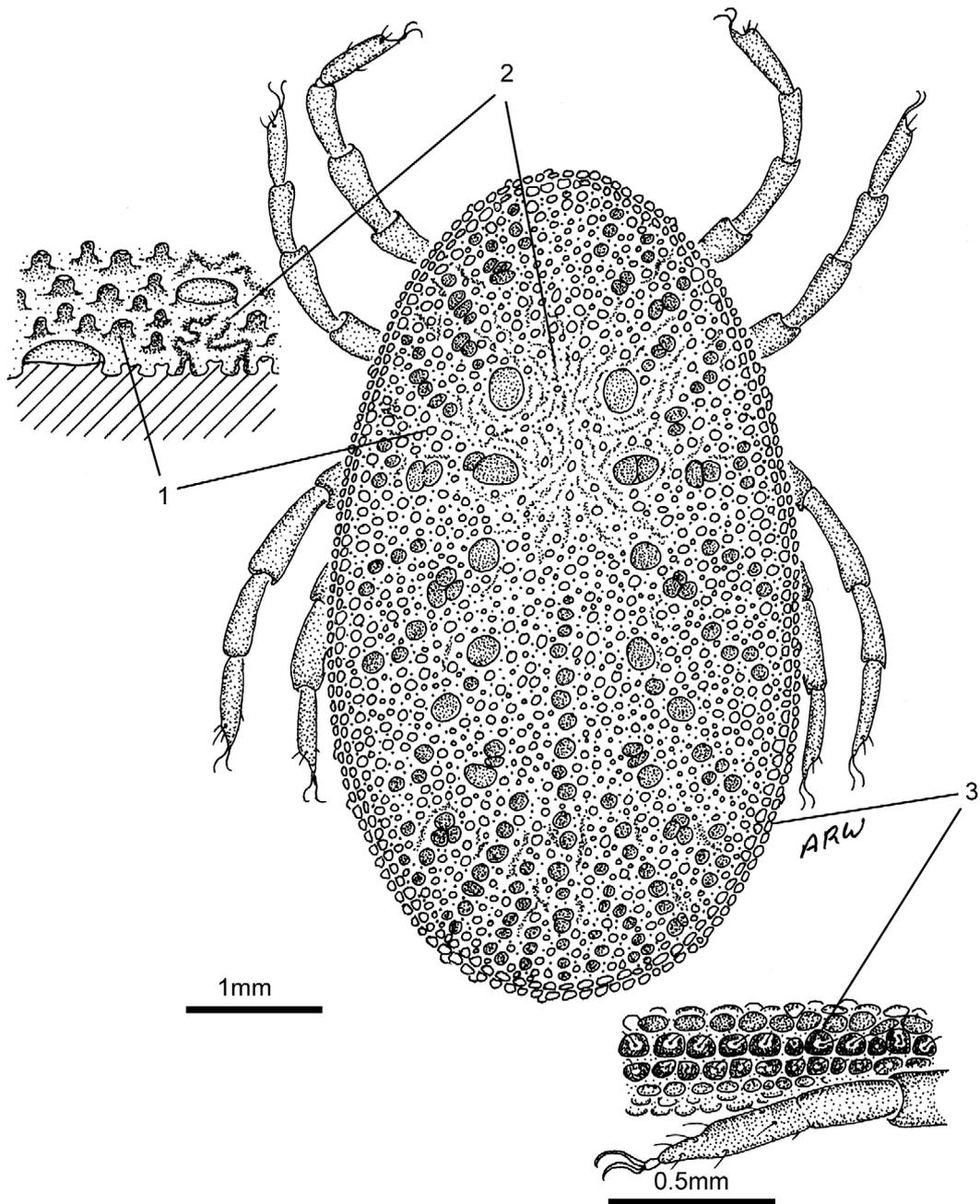
Argas robertsi is known as Robert's bird tick in Australia and elsewhere. *Argas robertsi* is a parasite of wild birds in Australia and elsewhere, but has been found also on domestic fowl in southwestern Queensland (Hoogstraal *et al.* 1974) and in Indonesia (see section on Hosts below). *Argas robertsi* was named after the great Australian tick taxonomist F.H.S. Roberts of Brisbane, Australia (see the introduction to this book for notes on F.H.S. Roberts.).

Differential diagnosis

See Figs 21 and 22. This is an abbreviated version of the differential diagnosis for *A. persicus*. Adults of *A. robertsi* measure: 6.0 mm by 3.5 mm, whereas adults of *A. persicus* measure 7.0 mm by 4.5 mm. For integumental patterns: *A. robertsi* has common mammillae but the ridges are sparse, also the mammillae in *A. robertsi* at the lateral suture are small whereas *A. persicus* has sparse mammillae but ridges are common; also the mammillae on *A. persicus* at the lateral suture are large. Both *A. robertsi* and *A. persicus* have mouthparts characterised by pairs of post-hypostomal setae and post-palpal setae.

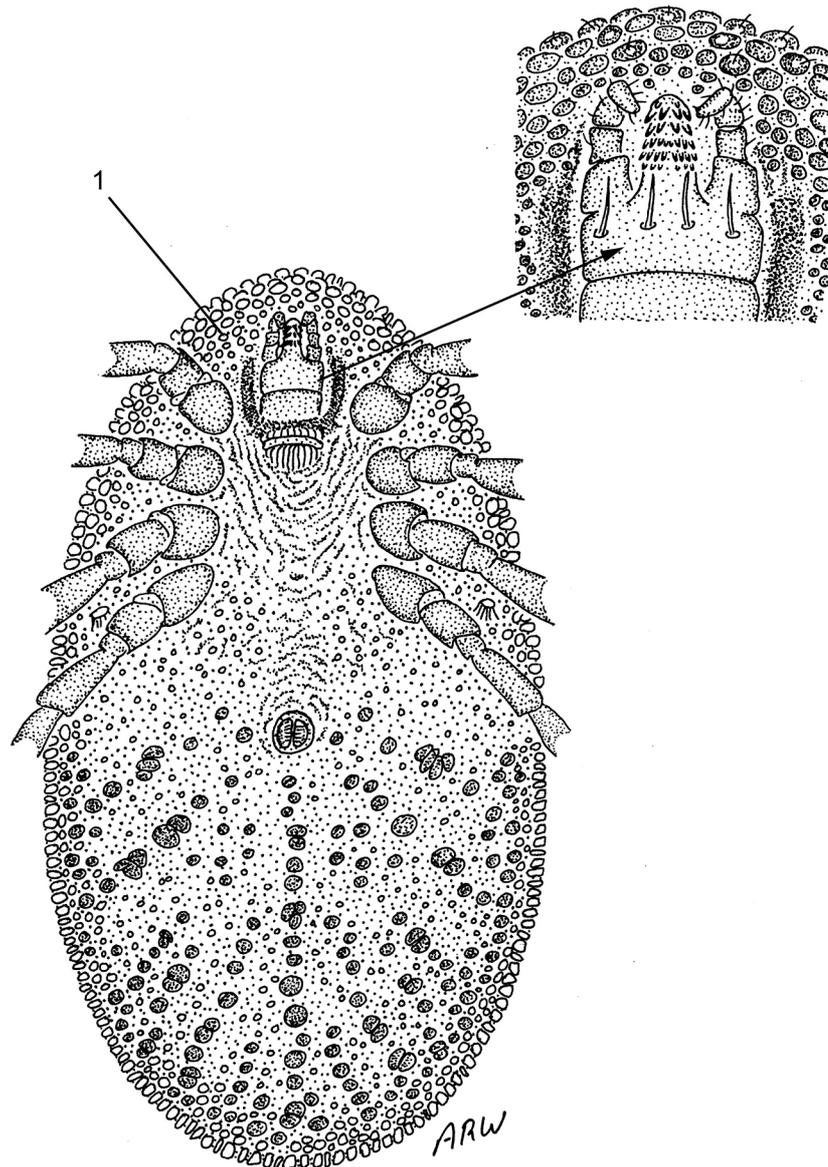
Hosts

Argas robertsi is a parasite of egrets, cormorants, herons, storks, ibises and other wading birds that nest in rookeries (colonies), and domestic fowl when these wild birds visit or live in the vicinity of fowl-houses (Hoogstraal *et al.* 1974; Munaf *et al.* 1980). So far, *A. robertsi* has been found in fowl-houses and on domestic fowl only in southwestern Queensland and at Lake Cowal, NSW (Hoogstraal *et al.* 1974), and in Indonesia (Munaf *et al.* 1980, precise locality not given but two of the authors worked at Bogor on the island of Java so it is likely that Bogor was the locality).



- 1 Mammillae on integument are common.
- 2 Ridges on integument are sparse.
- 3 Mammillae at lateral suture are small.

FIGURE 21. *Argas robertsi* (Robert's bird tick), female, dorsal.



1 Mammillae on integument are common.

FIGURE 22. *Argas robertsi* (Robert's bird tick), female, ventral.

Australian host records (from two different sources): (i) *Gallus gallus* (domestic fowl, introduced) (Roberts 1970); and (ii) *Phalacrocorax carbo novaehollandiae* (great cormorant) Lake Cowal, New South Wales; domestic fowl-houses, Lake Cowal; and nests in tree-holes of *Eolophus roseicapillus* (galah), Lake Cowal (Hoogstraal *et al.* 1974).

Host-records from outside of Australia (from three different sources): (i) *Pelecanus philippensis* (grey pelican); *Phalacrocorax carbo novaehollandiae* (great cormorant); *P. pygmeus* (pygmy cormorant); *P. sulcirostris* (little black cormorant); *P. melanoleucos* (little pied cormorant); *P. niger* (little cormorant); *Anhinga melanogaster* (oriental darter); *Ardea cinerea* (grey heron); *A. ibis* (cattle egret); *A. purpurea* (Africa heron); *A. picata* (pied heron); *Ardeola speciosa* (Javan pond heron); *Egretta garzetta* (little egret); *Ardea alba* (great egret); *A. intermedia* (intermediate egret); *Nycticorax nycticorax* (black-crowned night heron); *Anastomus oscitans* (Asian open billed stork); *Threskiornis melanocephalus* (oriental white ibis); *T. molucca* (Australian white ibis); *Plegadis falcinellus*

(glossy ibis); *Dendrocygna javanica* (lesser whistling teal or tree duck); *Tadorna ferruginea* (ruddy shelduck or Brahminy duck); *Anas crecca* (Eurasian or common teal); *A. querquedula* (garganey); *Eudynamis scolopacea* (Asian koel); *Otus bakkamoena* (Indian scops owl); *Athene brama* (spotted owl); *Gracupica contra* (pied myna); *Acridotheres javanicus* (Javan myna); *Sturnus nigricollis* (black-collared myna); and *Corvus macrorhynchos* (jungle crow) (Hoogstraal *et al.* 1974); (ii) domestic fowl (*Gallus gallus*) in Indonesia (Munaf *et al.* 1980); and (iii) rookery of an unknown wading bird on the island of Pulau Dua, north of Banten in the Java Sea (Wilson 1970, record BBM-Isa 85060).

Life-cycle and seasonality

The life-cycle of *A. robertsi* is typical of argasid ticks. Hoogstraal *et al.* (1975) described the life-cycle of *A. robertsi*, which had been collected in Australia, in the laboratory at 28–30° C and 75% relative humidity; ticks were fed on domestic pigeons. Eggs hatched after 14–29 days. Larvae attached to the host 3–8 days after hatching and fed to repletion in 4–10 days. There were 2–5 nymphal instars. Nymphs fed a few days after moulting. Nymphs fed to repletion in 23–42 minutes. Coxal fluid was usually secreted, as one or two drops, during feeding but on rare occasions after the nymphs had detached. Adult females were ready to feed 1–16 days after moulting from nymphs. About half (47 %) of the female ticks did not oviposit after their first meal; these females needed a second meal 1 to 2.5 months after the first meal. Then these females oviposited 4–16 days later. In the laboratory, the life-cycle of *A. robertsi* from Australia took 6.5 months in optimal conditions; it doubtless takes longer in nature. Hoogstraal *et al.* (1975) reported substantial variation in the life-cycles of *A. robertsi* from Australia (Adelaide River heron rookery, near Darwin, Northern Territory), Taiwan, Thailand, Indonesia and Sri Lanka.

Argas robertsi may be found throughout the year under the bark of trees that contain nests of cattle egrets, *Ardea ibis*, in southern Queensland (Standfast *et al.* 1987). Thus, future workers in Australia and elsewhere might look for *A. robertsi* under the bark of trees in the rookeries of other cattle egrets, other egrets, herons, storks, ibises and other wading birds.

Disease

Argas robertsi in Australia may harbour at least five arboviruses: (i) Lake Clarendon virus (= CSIRO 704) (St George *et al.* 1984) which was provisionally put in the family Reoviridae (Zeller *et al.* 1989). This virus was found in nymphs and adults of *A. robertsi* from a rookery (colony) of cattle egrets, *A. ibis*, at Lake Clarendon near Gatton in southern Queensland (St George *et al.* 1984). Humphrey-Smith *et al.* (1991a) reported antibodies to Lake Clarendon virus in seabirds on Heron Island and other islands of the Great Barrier Reef and Coral Sea, Queensland, Australia, but not in humans from Heron Island. Standfast *et al.* (1987) proposed that Lake Clarendon virus overwinters in *A. robertsi*. That is, Lake Clarendon virus persists from March to September (7 months) in the *A. robertsi* that live under the bark of trees that contain egret nests. Most of the cattle egrets in Queensland migrate to Victoria, Tasmania, South Australia and Western Australia for the winter-spring (March to September) (Pizzey & Knight (2010); (ii) CSIRO virus 1499, which, like Lake Clarendon virus, was first isolated from *A. robertsi* from the rookery of cattle egrets, *A. ibis*, at Lake Clarendon in southern Queensland. Humphrey-Smith *et al.* (1991a) reported antibodies to CSIRO virus 1499 in the sera of 1 of 101 (1 %) humans and 13 of 401 (3.2 %) *Anous minutus* (black noddies) from Heron Island on the Great Barrier Reef, Queensland. Experimental infection of cattle egrets, *A. ibis*, with CSIRO virus 1499 caused mortality in these birds (Standfast pers. comm. cited by Humphrey-Smith *et al.* 1987); (iii) Kao Shuan virus (Doherty *et al.* 1976). This virus was found in adults and nymphs of *A. robertsi* from the bird rookery near the Adelaide River, Northern Territory, that Khalil, Hoogstraal and Oliver studied (Khalil *et al.* 1980). Harry Hoogstraal personally collected those ticks (Doherty *et al.* 1976). Kao Shuan virus has also been found in *A. robertsi* from Taiwan and Java, Indonesia (Hoogstraal *et al.* 1974); (iv) Pathum Thani virus, a virus that is related to Kao Shuan virus (above) (Hoogstraal *et al.* 1974). Pathum Thani virus has been found in *A. robertsi* from a bird rookery near the Adelaide River, Northern Territory, Australia as well as from Java, Taiwan, Thailand and Sri Lanka; and (v) Nyamanini virus which has been found in *A. robertsi* from Thailand and Sri Lanka (Hoogstraal *et al.* 1974).

Argas robertsi may cause disease in cattle egrets. McKilligan (1987), in a seminal study, estimated the mortality of nestling cattle egrets due to *A. robertsi* to be 10.1 to 11.8 % from 1980–1982. McKilligan (1987) attributed this mortality to blood lost to ticks during tick feeding, possible toxic effects of tick-saliva, Lake Clarendon virus or some combination of these three.

Habitat and geographic distribution

The preferred climates and habitats of *A. robertsi* are not well defined but Hoogstraal *et al.* (1974) has detailed descriptions of the habitats and climates of the *A. robertsi* localities they studied in the Northern Territory (Australia), Indonesia, Thailand, India, Sri Lanka and Taiwan. So far, *A. robertsi* has been found in only two of the eight zoogeographical regions: Australasia and Indo-Malaya.

Argas robertsi has spread widely on birds like the cattle egret, *Ardea ibis*, and on other wading birds (see Hoogstraal *et al.* 1974). Whether or not *A. robertsi* has the same or a different geographic distribution in Australia as that of *A. persicus* is unknown. Roberts (1970) found only *A. persicus* in poultry-houses in Charleville, Winton, Blackall, Longreach and Dalby (Qld), Perth (WA), Adelaide (SA) and Moree, Narrabri and Dubbo (NSW). Petney *et al.* (2004) reported *A. robertsi* from a colony (rookery) of cattle egrets, *A. ibis*, in Queensland (locality not given). Roberts (1970) found that poultry-houses in Kalbar, Millmerran and Goondiwindi (Qld) had both *A. robertsi* and *A. persicus* (Roberts 1970). [Incidentally, the fact that *A. robertsi* was living along with *A. persicus* (i.e. living in sympatry) is evidence that *A. robertsi* and *A. persicus* are indeed different species]. Hoogstraal *et al.* (1975) reported *A. robertsi* from a light-house beacon on a small island in the Great Barrier Reef, Queensland, Australia; domestic poultry were not on this island. McKilligan (1987) reported that *A. robertsi* was absent from five cattle egret, *A. ibis*, rookeries (heronries) in southern Queensland; so it seems that *A. robertsi* has not yet infested all cattle egret colonies in Queensland. Future workers might record, in the scientific literature, which rookeries of the cattle egret, *A. ibis*, are infested and which are not infested so that the spread of *A. robertsi* might be better understood.

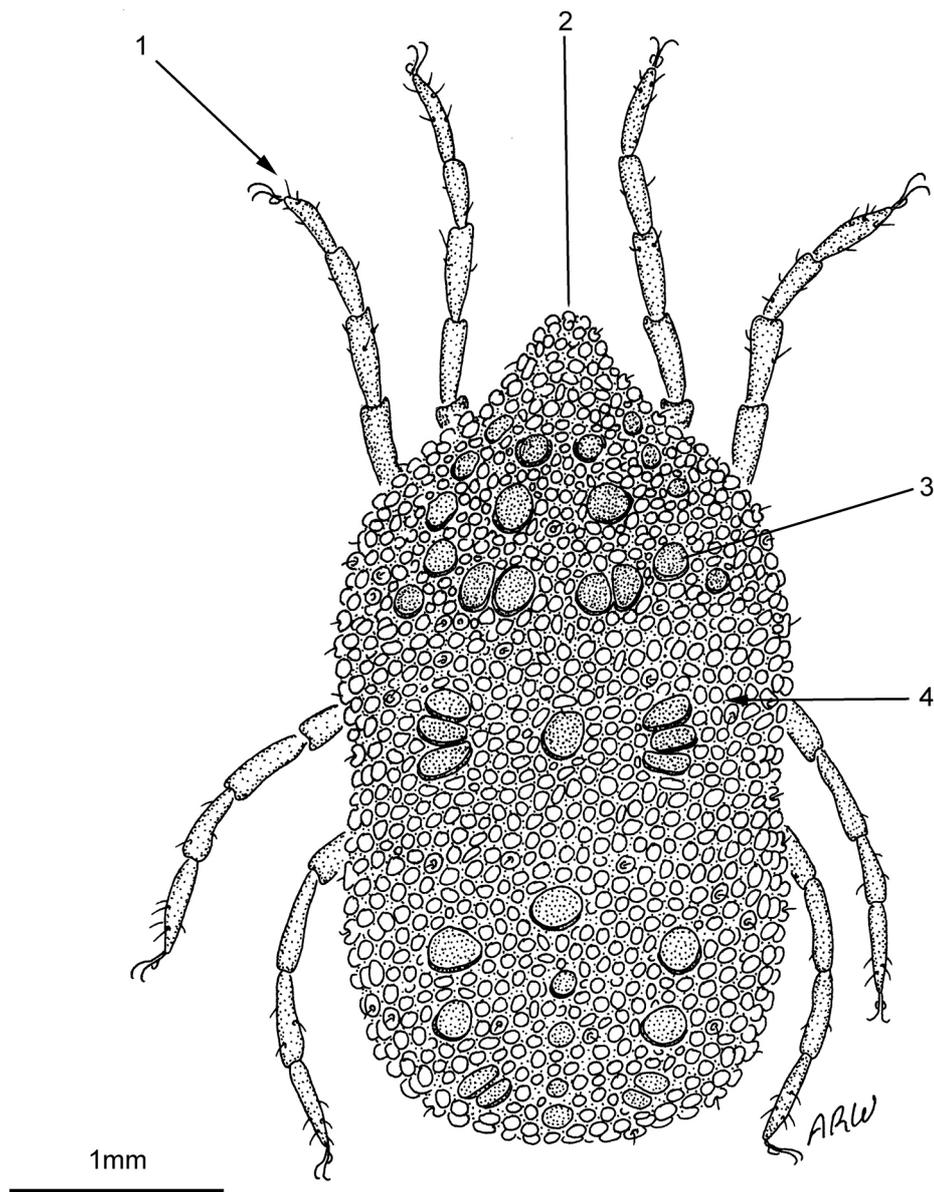
Genes and genomes

Part of only one gene is in GenBank and thus available to the public: the small subunit mitochondrial rRNA (16S) (398 bp, Petney *et al.* 2004).

Other information

Despite the fine work of Petney *et al.* (2004) and Kohls *et al.* (1970) there is still some uncertainty about the status of *A. robertsi* in Australia since the 16S rRNA (mitochondrial) nucleotide sequence in GenBank attributed to *A. robertsi* from a cattle egret, *A. ibis*, colony in Queensland, Australia (AY436768; Petney *et al.* 2004) is near-identical to the 16S sequence in GenBank attributed to *A. miniatus* from Brazil (mt genome number KC76B9590) (Burger *et al.* 2014a). These authors concluded that the most likely explanations for the near-identical 16S rRNA nucleotide sequences of *A. robertsi* from Australia and *A. miniatus* from Brazil were either: (i) that *A. robertsi* and *A. miniatus* are regional variants of the same species; or (ii) that the 16S sequence of *A. robertsi* in GenBank (AY436768) from Queensland is actually from *A. miniatus* i.e. that *A. miniatus* might occur in Queensland. Further study of the genetics of *A. robertsi* (Australia and Asia) and *A. miniatus* (South America) is required to reject one or both of these hypotheses (Burger *et al.* 2014a). This uncertainty about the status of *A. robertsi* and *A. miniatus* echoes to us the opening statement of Kohls *et al.* (1970): "For many years much confusion has existed as to the poultry-infesting ticks of the subgenus *Persicargas* of the New World" (We interpret "New World" as three of the eight zoogeographical regions: Neotropics, Australasia and Oceania). Further genetic and morphological studies of the poultry-infesting *Argas* ticks of the New World will likely uncover as yet undescribed species of ticks. *Argas robertsi*, which so far is known only from the Australasian and Indomalay regions may be the sister-species ("counterpart" in the words of Hoogstraal *et al.* 1974) of *A.(P.) arboreus* of the Afrotropical zoogeographical region (Hoogstraal *et al.* 1974). Indeed, *A. robertsi* and *A. arboreus* interbreed readily and produced partially fertile progeny in the laboratory (Khalil *et al.* 1980). These authors hypothesised that *A. robertsi* and *A. arboreus* have a MRCA (i.e. Most Recent Common Ancestor) and that *A. robertsi* and *A. arboreus* evolved by allopatric speciation.

Ornithodoros capensis Neumann, 1901 (seabird soft tick)



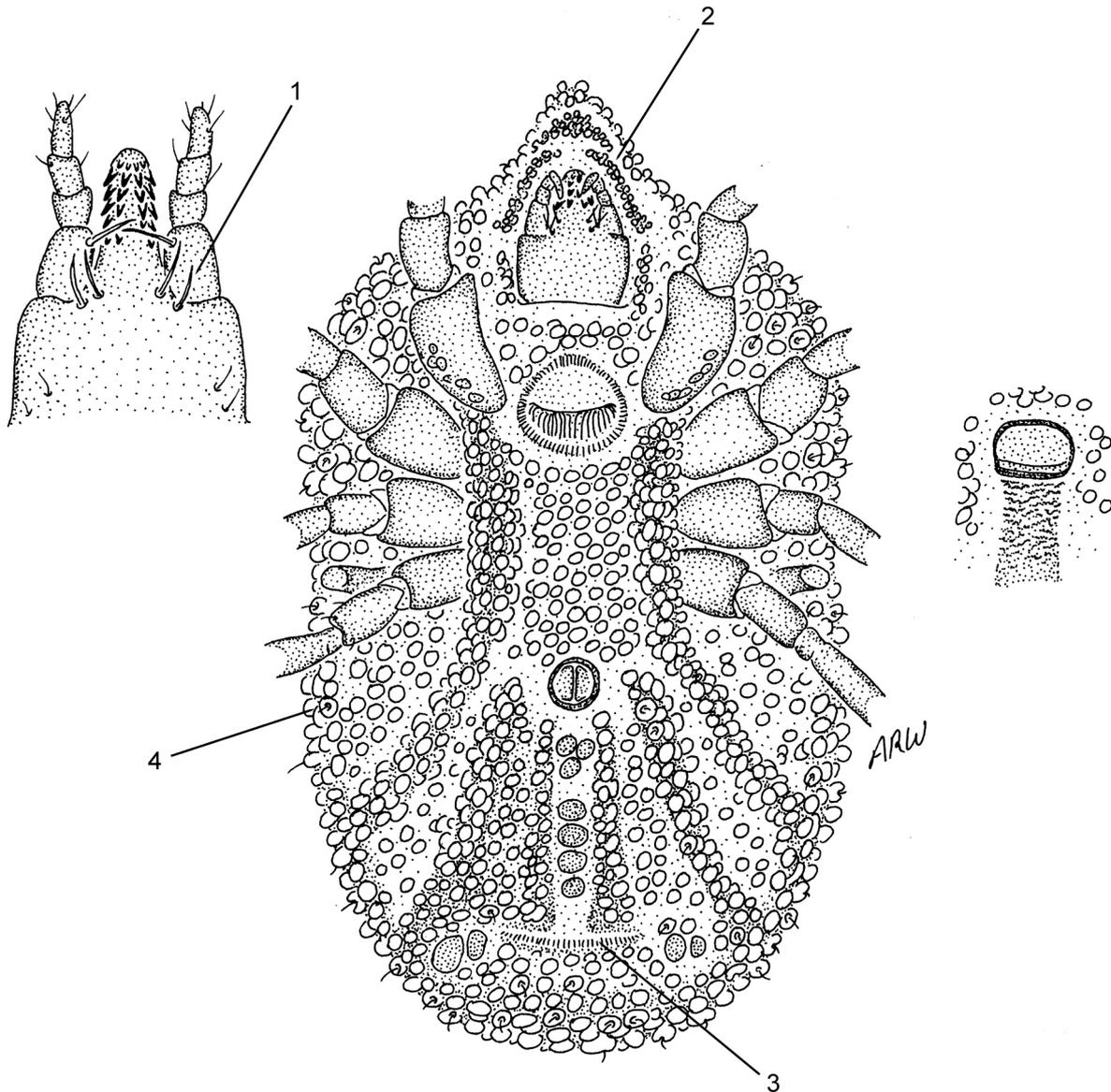
- 1 Tarsal dorsal humps are absent.
- 2 Anterior projection of body is present.
- 3 Discs on main body are present.
- 4 Ridged areas on main body are absent.

FIGURE 23. *Ornithodoros capensis* (seabird soft tick), female, dorsal.

General.

In Australia and elsewhere, *Ornithodoros capensis* is known as the seabird soft tick. *Ornithodoros capensis* is one of 10 species in the *O. capensis* group of species of the subgenus *Alectorobius* (Hoogstraal 1985). Most of the other 36 species in the subgenus *Alectorobius* live in the Neotropical zoogeographic region (see Camicas *et al.* 1998). Eight of the 10 species in the *O. capensis* group infest seabirds; indeed seabirds of at least 12 families. In Australia, humans are most likely to encounter *O. capensis* on off-shore islands, particularly coral cays, as campers,

fisherman or explorers. Note that *O. capensis* has been known as *Carios capensis* in the past. See Burger *et al.* (2014a, Table 1), Guglielmo *et al.* (2010) and Estrada-Peña *et al.* (2010) for a discussion of the tumult in classification of the genus *Ornithodoros* and the other genera of soft ticks.



- 1 Postpalpal setae are present.
- 2 Camerostome cheeks consist of a single flap on each side.
- 3 Transverse postanal groove is indistinct and single.
- 4 Setae are sparse on dorsal and ventral surfaces.

Note: genital aperture of an adult male is shown to same scale, inset top right.

FIGURE 24. *Ornithodoros capensis* (seabird soft tick), female, ventral.

Differential diagnosis

See Figs 10B, 23 and 24. *Ornithodoros capensis* is the only species of *Ornithodoros* known to bite humans in the vicinity of seabirds and sea bird nests, so differential diagnosis is usually straight-forward. To differentiate *O.*

capensis from *O. gurneyi*, the only other species of *Ornithodoros* considered in this book, the most conspicuous characters for *O. capensis* are the small size and pale colour, absence of humps on the tarsi of the legs, and presence of discs on the dorsal surface of both sexes. *Ornithodoros gurneyi* in comparison is large and dark, with tarsal dorsal humps and without discs. Less distinct differences can be found on the ventral surfaces. *Ornithodoros capensis* has postpalpal setae, camerostome cheeks of a single flap on each side, and a single postanal groove whilst *O. gurneyi* has no postpalpal setae, multiple flaps of the camerostome cheeks, and paired postanal grooves. Beyond Australia, *O. capensis* has been confused with *O. coniceps* (Canestrini, 1890) since these two species are morphologically very similar. *Ornithodoros coniceps*, however, lives in very humid, rocky, mainland habitats and feeds on swallows, swifts, sparrows, doves and even domestic pigeons and poultry (Hoogstraal *et al.* 1979). Also, in adult and nymphal *O. capensis*, the rows of integumental mammillae are evenly and closely spaced compared to the rows of integumental mammillae of *O. coniceps* which are in chain-like rows (Hoogstraal *et al.* 1976, 1979).

Hosts

Ornithodoros capensis is a tick of seabirds, particularly terns, gulls and penguins, that will feed on humans and domestic fowl given the opportunity (Kohls *et al.* 1965).

Australian host records: (i) *Eudyptula minor* (little penguin); *Sula leucogaster* (brown booby); *Sterna fuscata* (sooty tern); *Anous minutus* (black noddy); (Roberts 1970). Roberts (1970) did not give any specific information on his host records of *Su. leucogaster*; *St. fuscata* nor *A. minutus* so new records of these hosts being infested with *O. capensis* would be welcome in the scientific literature. In a similar vein, Roberts (1970) did not give specific records of *O. capensis* on *E. minor*. Rather, he referred to the "various localities along the coast from Perth in Western Australia to Sydney in New South Wales with *E. minor*" cited by Kohls *et al.* (1965). Curiously, the one study of the ticks of *E. minor* did not reveal a single *O. capensis* (Mykytowycz & Hesterman 1957). These authors handled about 100 birds, mostly chicks, "examined carefully" 50 chicks and eight adults, and examined nest material, on Montagu and Gabo Islands, New South Wales. Nine of the 50 chicks and two of the adult birds had *Ixodes kohlsi* but no *O. capensis*. *Ixodes kohlsi* was also found in the nest material but *O. capensis* was not found. Nonetheless, there is a record of *O. capensis* from the nest of *E. minor* on Lady Julia Percy Island (Tubb, 1937); a record of *O. capensis* from *E. minor* somewhere in Western Australia (Ferguson, 1925); and a record of *O. capensis* from *E. minor* on Lion Island, New South Wales (Kohls *et al.* 1965). The host of the single female *O. capensis* collected on English Island, Sir Joseph Banks Group, Spencer Gulf, South Australia, might have been *E. minor* but this needs to be confirmed before it could be accepted as a host-record. In a similar way, the host of the adult and nymphs, near penguin nests (RML 30721, Five Island, off Point Kembla, NSW Mar. 11, 1946, H. C. Davis) cited by Kohls (1957) needs to be confirmed, but may have been *E. minor* since this is the most likely species of penguin to be nesting in New South Wales. The host record "several larvae, probably this species [*O. capensis*], ex legs of the burrowing owl, *Speotyto cunicularia rostrata*, (RML, 22 326, Vivionne Bay, Kangaroo Island, South Australia, Australia, Jan 24, 1946 H. Womersley)" is rejected by us since *Athene* (= *Speotyto*) *cunicularia* has not been recorded in Australia (Pizzey & Knight 2010) plus we are dubious about the identification ("several larvae probably [*O. capensis*]"); (ii) *A. minutus* (black noddy), humans (Humphrey-Smith *et al.* 1991b); and (iii) ten nests of *Anous minutus* (black noddy) and from humans, Heron Island, Great Barrier Reef, Coral Sea, Queensland (Barker and Campelo unpubl. data).

Host-records from outside Australia (from five different papers): (i) *Stictocarbo punctatus punctatus* (spotted shag) (Dumbleton, 1961); (ii) *Spheniscus demersus* (Jackass penguin), *Actitis macularia* (spotted sandpiper), *Sula leucogaster* (brown booby), *Platalea ajaja* (roseate spoonbill), *Anous stolidus* (brown noddy), *Phoebastria immutabilis* (Laysan albatross), *Onychoprion fuscatus* (sooty tern), *Eudyptula minor* (little penguin), and *Phalacrocorax punctatus* (spotted shag) (Kohls *et al.* 1965); (iii) *Diomedea nigripes* (black footed albatross), *Phoebastria immutabilis* (Laysan albatross), *Ardenna pacificus* (wedge-tailed shearwater), *Phaethon rubricauda* (red-tailed tropic bird), *Sula dactylatra* (masked booby), *S. leucogaster* (brown booby), *S. sula* (red-footed booby), *Fregatta ariel* (lesser frigatebird), *Arenaria interpres* (ruddy turnstone), *Sterna lunata* (spectacled tern), *S. fuscata* (sooty tern), *Procelsterna cerulea* (blue noddy), *Anous stolidus* (brown noddy), *Anous tenuirostris* (lesser noddy), and *Gygis alba* (white tern) (Amerson 1968); (iv) *Larus novaehollandiae* (silver gull) (ticks were under rocks in a rookery) (Heath 2006); and (v) *Pterodroma neglecta* (Kermadec petrel), *P. nigripennis* (black-winged petrel), *Puffinus assimilis* (little shearwater), *Phaethon rubricauda* (red-tailed tropicbird), *Morus serrator* (Australasian gannet), *Stictocarbo punctatus punctatus* (spotted shag), *Chroicocephalus novaehollandiae scopulinus* (red-billed

gull), *Anous stolidus pileatus* (common noddy), *Onychoprion fuscatus serratus* (sooty tern) and the land bird *Porzana tabuensis tabuensis* (spotless crane) (Heath *et al.* 2011).

Life-cycle and seasonality

The life-cycle of *O. capensis* is apparently typical of argasid ticks, although we note that the life-cycle has not yet been studied entirely in the laboratory. Heath (2006) found that wild caught *O. capensis* fed readily on domestic fowl. Thus, domestic fowl might be used by future workers to describe, entirely, the life-cycle of *O. capensis*. Male and female *O. capensis* copulated in a glass vial shortly after feeding; a spermatophore was left in the genital aperture of the female (Heath 2006). Females first laid eggs 45 to 108 days after feeding. Eggs hatched after about 31 days. Wild-caught nymphs (unknown instar) moulted to the next nymphal instar 32 to 115 days after feeding. Subsequent instars moulted to the next instar up to 76 days after feeding (Heath 2006). All of these experiments were apparently at room temperature.

Disease

Ornithodoros capensis is notorious for the viruses that have been isolated from it (Hughes *et al.* 1964, Doherty *et al.* 1969, Doherty 1972, Converse *et al.* 1975, Austin 1978) and *Rickettsia* spp. (Reeves *et al.* 2006, Kawabata *et al.* 2006, Mattila *et al.* 2007, Duh *et al.* 2010). Yet we cannot find a single confirmed case of disease in humans caused by one of these viruses or a *Rickettsia* species. Humans may even be serum-positive for some of these viruses yet those people were seemingly well (Humphrey-Smith *et al.* 1987). Nonetheless, St George *et al.* (1977) were drawn to reports of illness in people who built and serviced the automatic weather reporting stations on Saumarez and Frederick Reefs, and other coral cays of the Great Barrier Reef, Queensland, Australia. These people reported being attacked by large numbers of ticks, probably *O. capensis*, some of which attached. An unknown number of these people developed febrile illness, but alas, their serum was not tested for viruses. Votyakov (1974), however, associated illness in people with Tyuleniy virus which might have been transmitted to people by *O. capensis* but we were unable to get a copy of Votyakov (1974) which was cited by St George *et al.* (1977). These patients had chills, malaise, fever, headaches, joint pains, enlarged lymph nodes and haemorrhagic rashes on their limbs and trunk (Votyakov 1974). The incubation period was 1–8 days; the patients recovered completely.

Whereas concrete evidence of disease caused by viruses and a *Rickettsia* species transmitted by *O. capensis* to humans is lacking in Australia, there is no doubt that people who live on coral cays of the Great Barrier Reef, for example, Heron Island, Queensland may develop severe hypersensitivity to the bites of larval, nymphal and adult *O. capensis*. For example, see the photographs of the forearms of people who lived and worked on Heron Island in Humphrey-Smith *et al.* (1991b Fig. 1). At least three people personally known to one of us (SB) became highly sensitized to the bites of larval, nymphal and adult *O. capensis* while living and working on Heron Island, Queensland. Humphrey-Smith *et al.* (1991b) reported intense pruritus, blistering (a major feature), erythema, weeping, lesions, lymphangitis, dull ache, rheumatic pain, and general lassitude and discomfort in response to what was believed to be the bites of *O. capensis*.

Ornithodoros capensis may also have a detrimental effect on its bird hosts. King *et al.* (1977) reported that heavy infestations of *O. capensis* caused *Pelecanus occidentalis* (brown pelican), in Texas, USA, to desert their nests and abandon their colony. Amerson (1966) found nymphal *O. capensis* in the nasal cavities of 2–3 week old chicks of *Sterna fuscata* (sooty tern), however, disease was not associated with these nasal infestations. Conceivably, viruses and *Rickettsia* species transmitted among seabirds by *O. capensis* may have caused disease in these birds but this has not yet been investigated, but Morgan *et al.* (1985) reported mortality in *Eudyptula minor* (little penguin) after experimental infection with Saumarez Reef virus.

Habitat and geographic distribution

Ornithodoros capensis is a tick of the nests of seabirds that breed on islands and occasionally on mainlands. As yet, there are few hard data on the geographic distribution of *O. capensis* in Australia. New locality records (and host records) would be welcome additions to the scientific literature.

Australian localities from Roberts (1970). Roberts (1970) did not give specific localities but rather referred only to the various localities along the coast from Perth in Western Australia to Sydney in New South Wales, with *Eudyptula minor* (little penguin) as host cited by Kohls *et al.* (1965). Roberts (1970) also reported that *O. capensis* was present on "certain atolls" of the Coral Sea in association with *Anous stolidus* (brown noddy), *Sula leucogaster* (brown booby) and *Sterna fuscata* (sooty tern).

World localities: *Ornithodoros capensis* lives on the islands and occasionally, on the mainlands of the tropics and subtropics of the Pacific, Atlantic and Indian Oceans between 40°N and 45°S (Heath *et al.* 2011).

Genes and genomes

Parts of the following genes are available to the public in GenBank: (i) nuclear small subunit (18S) and large subunit (28S) rRNA (ca. 267 bp and 216 bp, Crampton *et al.* 1996); (ii) mitochondrial small subunit (16S) rRNA (ca. 475 bp, Ushijima *et al.* 2003); (iii) mitochondrial large subunit (16S) rRNA (ca. 423 bp, Kawabata *et al.* 2006; ca. 424 bp Gomez-Diaz *et al.* 2012); (iv) nuclear large subunit (18S) ribosomal RNA (ca. 529 bp, Gomez-Diaz *et al.* (2012); and (v) the entire mitochondrial genome 14,418 bp (Shao *et al.* 2004).

Other information

None.

Ornithodoros gurneyi Warburton, 1926 (kangaroo soft tick)

General

Ornithodoros gurneyi is known as the kangaroo soft tick. *Ornithodoros gurneyi* is one of four species in the subgenus *Ornamentum* (Camicas *et al.* 1998). Curiously, the other three species in this subgenus live in the Neotropical zoogeographical region and in the case of one species, *O. coriaceus*, in the Neotropical and Palearctic region. Only three of the 112 species of *Ornithodoros* are known from Australia: *O. gurneyi* from kangaroos, *O. capensis* from seabirds and *O. macmillani* from tree hollows.

Differential diagnosis

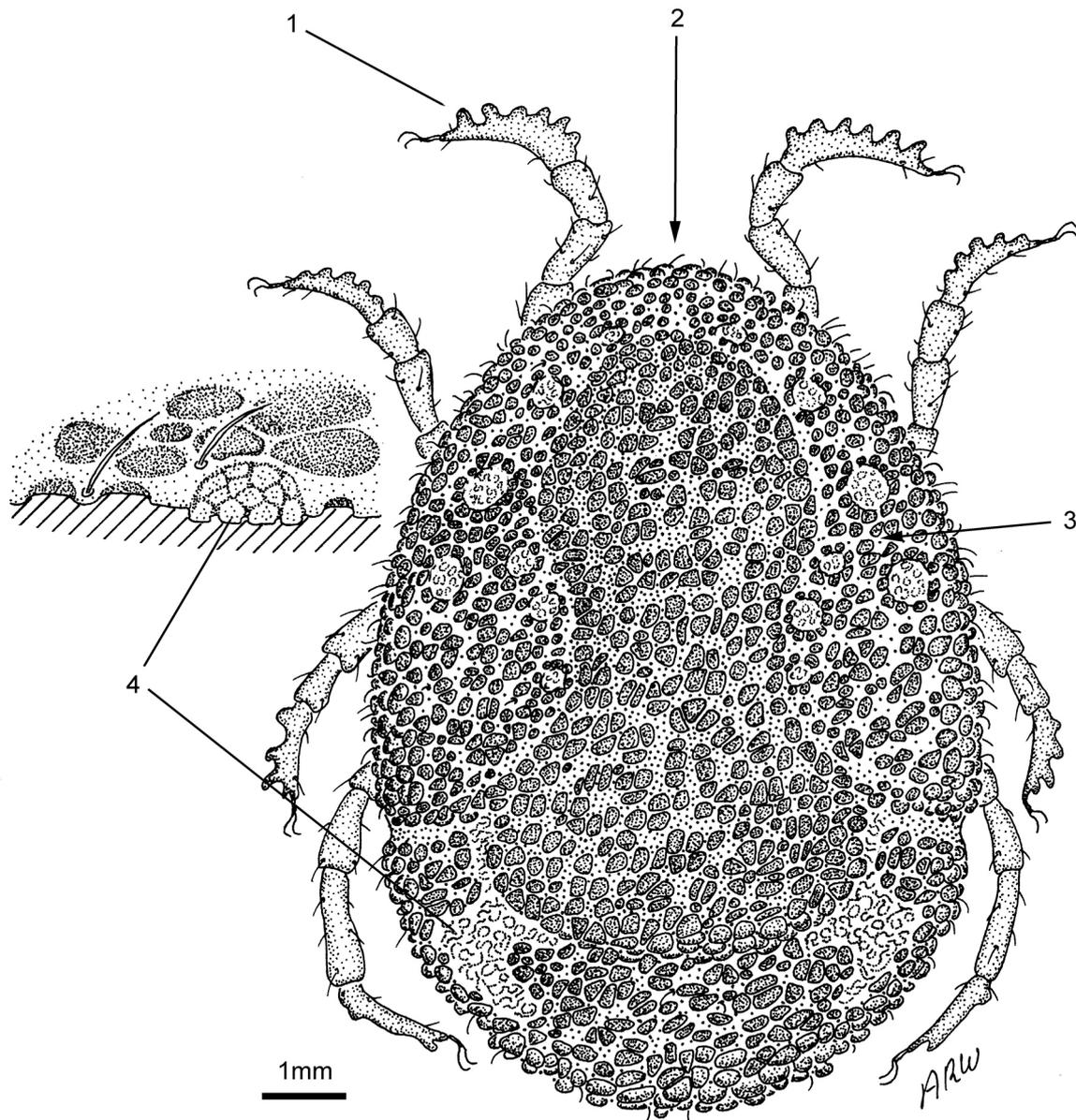
See Figs 25 and 26. *Ornithodoros gurneyi* is the only species of *Ornithodoros* known to bite domestic animals in Australia so differential diagnosis is usually straight-forward. *Ornithodoros gurneyi* and/or *O. capensis* (which may bite humans) of both sexes are easily differentiated using morphological features. *Ornithodoros gurneyi* is large and dark, with tarsal dorsal humps and without discs. For *O. capensis*, the most conspicuous differences are the small size and pale colour, absence of humps on the tarsi of the legs, and presence of discs on the dorsal surface. Ventrally, less distinct differences can be found. *Ornithodoros gurneyi* has no postpalpal setae, camerostome cheeks with multiple flaps on each side, and a paired postanal groove whilst *O. capensis* has postpalpal setae, single flaps of the camerostome cheeks, and a single postanal groove.

Hosts

Curiously, *O. gurneyi* has not yet been collected from a single native mammalian host! *Ornithodoros gurneyi* is a tick of the soil of "roo camps", the resting day-time places of kangaroos, principally *Macropus rufus*, the red kangaroo which is the symbol of Outback Australia, and *M. robustus* the common wallaroo. *Ornithodoros gurneyi* has, however, been collected from the skin of humans, domestic dogs and cattle and even *Pogona barbata*, the bearded dragon (Doube 1975a). We expect that the larvae of *O. gurneyi* will indeed be collected from native hosts, principally *M. rufus* and *M. robustus*, by future workers who are mindful of the small size of larval *O. gurneyi*. *Ornithodoros gurneyi* attached to rats in the laboratory for three days so we expect that these larvae attach to their native hosts for days rather than hours, too (Browning 1962, Doube 1975a). That *O. gurneyi* has fed on all laboratory animals tested thus far (rats, rabbits, mice, dogs, kangaroos and even humans, Doube 1975a), indicates that the specificity of *O. gurneyi* for *M. rufus* and *M. robustus* camps is due to the almost complete lack of opportunity of this tick to infest hosts other than *M. rufus* on the plains of central Australia and *M. robustus* on the hills.

Host (and habitat) records from Roberts (1952, 1970): habitat of *Macropus giganteus* (eastern grey kangaroo), *M. robustus* (common wallaroo), *M. rufus* (red kangaroo), humans, *Canis familiaris* (domestic dog, introduced), *Bos taurus* (domestic cattle, introduced) and *Equus caballus* (domestic horse, introduced).

Host records since Roberts (1970): habitat of *Macropus rufus* (red kangaroo) and *M. robustus* (common wallaroo) (Doube 1975c).



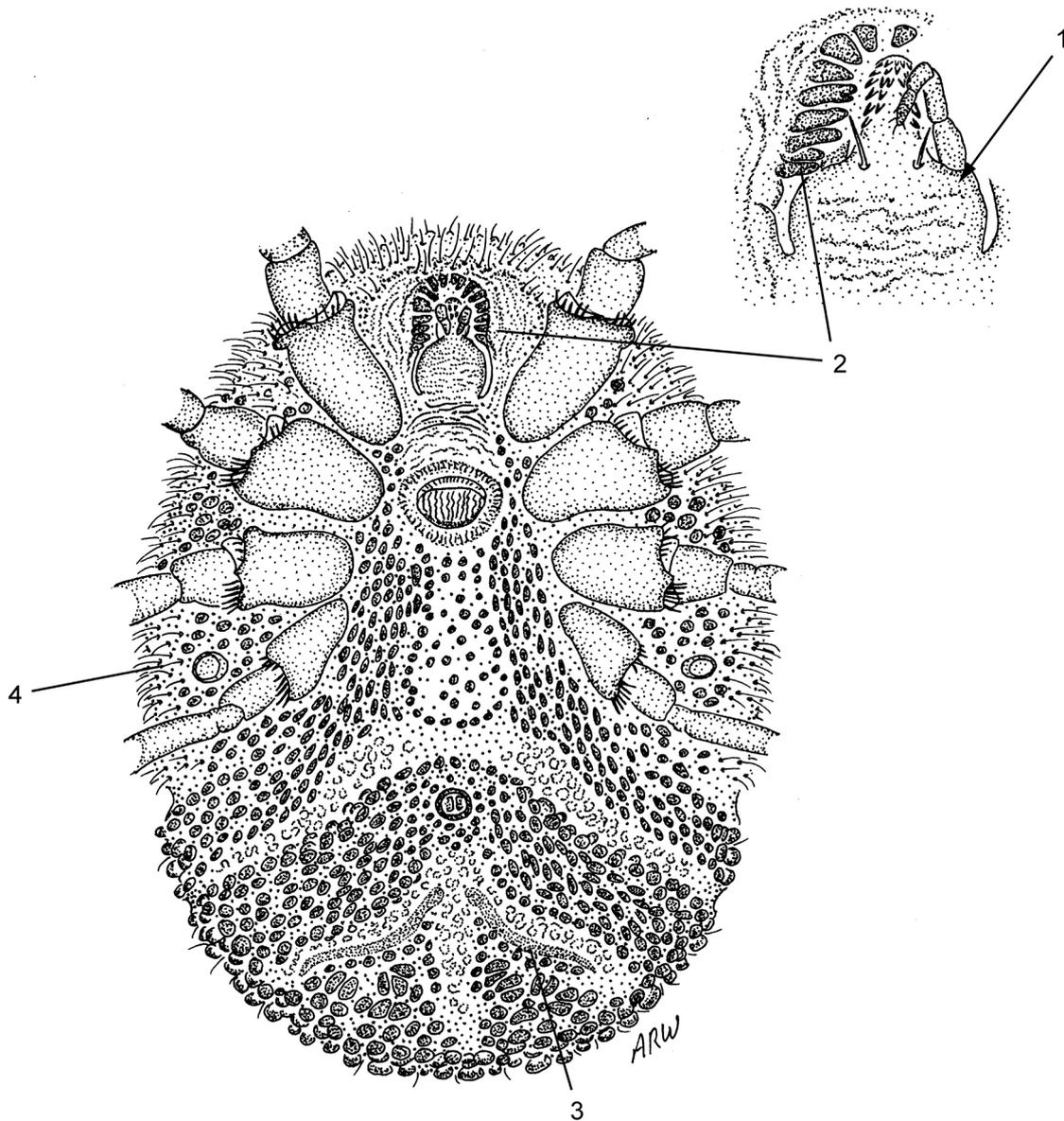
- 1 Tarsal dorsal humps are present.
- 2 Anterior projection of body is absent.
- 3 Discs on main body are absent.
- 4 Ridged areas on main body are present.

FIGURE 25. *Ornithodoros gurneyi* (kangaroo soft tick), female, dorsal.

Life-cycle and seasonality

The life-cycle of *O. gurneyi* is typical of argasid ticks. The life-cycle stages are eggs, larvae, three, four or five nymphal instars and adults (separate sexes). All of these life stages, except the eggs, apparently feed on kangaroos, particularly *Macropus rufus* on the plains and *M. robustus* on the hills of central Australia (Doube 1975a). Mating, either before or after females engorge, is in the soft soil of the wallows of *M. rufus*, underneath small trees, and in the wallows of *M. robustus* in caves where these kangaroos shelter from the sun (Doube 1972, 1975a). Female *O. gurneyi* lay up to six batches of eggs; each batch is 200 to 800 eggs. Often the female lies on top of her egg mass.

When the eggs hatch, the larvae may remain under the female tick in a large ball until she is disturbed and even then the larvae may cling to the females and/or hide in the crevices of her wrinkled body (Doube 1975a). Larvae usually attached to their host for four to six days (maximum 12 days). The engorged larvae then detach and return to the soil of the kangaroo wallow where they moult to first instar nymphs. There are three, four or five nymphal instars. Each nymphal instar feeds and then returns to the soil of the kangaroo wallow. Nymphs attach to their host for a much shorter period than the larvae; 20 minutes to 3 hours for nymphs compared to four to six days for larvae (Doube 1975a). Engorged larvae, and first and second instar nymphs, detach from their hosts in the daylight hours when their host is resting. Thus, ticks drop into kangaroo wallows rather than on the open plains (Doube 1975b). This detachment and behaviour is synchronised by circadian rhythms that can be entrained by the light regime (Doube 1975b).



- 1 Postpalpal setae are absent.
- 2 Camerostome cheeks consist of multiple flaps on each side.
- 3 Transverse postanal groove is distinct and paired.
- 4 Setae are dense on dorsal and ventral surfaces.

FIGURE 26. *Ornithodoros gurneyi* (kangaroo soft tick), female, ventral.

In the laboratory, at 30°C, one generation of *O. gurneyi*, with four nymphal instars, took about four months; but in nature, the time needed for one generation varies substantially since nymphs and adults enter reproductive diapause from midsummer (December) to midwinter (June) (Doube 1972, 1975d). The reproductive diapause is regulated by both temperature and photo-period. At 40°C, both nymphs and adults went into diapause. Diapause at high temperatures, such as 40°C, is doubtless an adaptation to life in the hot deserts of central Australia (Doube 1975d). Take for example, an engorged female; there would be no point laying eggs at 40°C since the larvae, should they emerge from the eggs, would die quickly. Diapause in *O. gurneyi* is also switched on when the light to dark ratio is changed from 8 hours dark: 16 hours light to 8 hours light: 16 hours dark (Doube 1975d). There is no obvious adaptive value to going into diapause when days get shorter so Doube (1975d) proposed that this was an evolutionary relict of a former life style, although it seems to us that this diapause may be adaptation to winter.

Habitat and geographic distribution

Ornithodoros gurneyi is one of the few exclusively desert-dwelling ticks of Australia. Like most species of *Ornithodoros*, *O. gurneyi* is a nest-dwelling tick. The nest of *O. gurneyi* is typically in the soft soil of the wallows of two species of kangaroos: *Macropus rufus* of the open plains and *M. robustus* in the hills of inland Australia (Doube 1972, 1975a). *Ornithodoros gurneyi* has also been taken from the burrows of unknown animals (mammals or birds we presume) in Malchi, Gracemere (near Rockhampton, Queensland) and from St Lucia, Brisbane, Queensland (Wilkinson 1958).

Ornithodoros gurneyi has only been collected from the following localities; doubtless it is more common than this small number of localities indicates. *Queensland localities*: Charters Towers (Roberts 1952); Lawn Hill, Julia Creek, Charters Towers, Mt Isa (Roberts unpublished data 1949 cited in Seddon 1968); Hughenden and Stamford (Qld) (Seddon 1968); Mt Isa (Doube 1975c); Malchi, Gracemere via Rockhampton, curiously in burrows of an unknown animal (Wilkinson 1958). *Western Australia localities*: Mileura Station, Cue Shire (Doube 1975c). *New South Wales localities*: Tibooburra (Henry 1938); Pine Creek via Broken Hill, Fowlers Gap (Doube 1975c). *South Australia localities*: Flinders Ranges, Moralana Station, and Kingoonya (Doube 1975c). *Northern Territory localities*: Macarthur River area (Anon 1928 cited by Seddon 1968) [Also "Western" and "Central" Australia (Seddon; 1968)]; Reynold Ranges (200 km north of Alice Springs), MacDonnell Ranges, Alice Springs (Doube 1975c). Seddon (1968) hypothesised that the southern limit of the distribution of *O. gurneyi* was the line drawn through the southern boundaries of Milparinka and Wanaaring Districts (north western NSW). Roberts (1952) stated that the most easterly locality for *O. gurneyi* was Charters Towers 60 km from the coast.

Disease

Ornithodoros gurneyi rarely encounters humans or livestock since this tick lives primarily in the wallows of desert-dwelling kangaroos. Should a human, dog, horse or other mammal, however, shelter under a desert-tree or in a desert-cave, *O. gurneyi* may emerge from the soil and attack. *Ornithodoros gurneyi* engorges quickly (5 to 40 mins) then falls back to the soil (Seddon 1968). The bite of *O. gurneyi* causes severe local and systemic reactions in humans (Roberts 1936, Henry 1938). A swelling as "big as a hen's egg", headache, impaired vision, even temporary blindness, vomiting and collapse have been recorded in humans (Roberts 1936, 1970; Henry 1938). The bite of this tick should be avoided!

Pope & Carley (1956) speculated that *O. gurneyi* might be the vector of a new species of *Borrelia* they found in *Rattus villosissimus* (long-haired rat). This species of *Borrelia* was later named *Borrelia queenslandica* by Carley & Pope (1962). These authors attempted to transmit *B. queenslandica* to laboratory mice with three individual *O. gurneyi* without success but their experiments were inconclusive, so the hypothesis that *O. gurneyi* is a vector of *B. queenslandica* remains to be tested.

Ornithodoros gurneyi may also be a vector of *Coxiella burnetii* (Q fever) but this is not certain. Roberts (1952, p. 211) stated that *O. gurneyi* can successfully transmit *C. burnetii* under experimental conditions but does not cite published work nor unpublished data. It seems to us that Roberts (1952 p. 211) was probably referring to Smith (1942) who had acknowledged Roberts for provisionally identifying *O. gurneyi*. *Ornithodoros gurneyi* did not infect guinea pigs with *C. burnetii* when ticks infected with this bacterium fed on uninfected guinea pigs, but guinea pigs did become infected with *C. burnetii* when homogenate-extracts of infected *O. gurneyi* were inoculated into guinea pigs (Smith 1942).

Genes and genomes

There is at present only one nucleotide sequence publically available of *O. gurneyi*: 16S RNA (423 bp, Petney *et al.* 2004).

Other information

Ornithodoros gurneyi comprises two races: the "cave" race whose principal host is *Macropus robustus* and the "plains" race whose principal host is *M. rufus* (Doube 1975c). Wild caught *O. gurneyi* may be fed on laboratory rats, rabbits and mice and thus laboratory cultures are readily made and maintained (Doube 1975c). Feldman-Muhsam & Filshie (1976) described the spermatophores of *O. gurneyi*.

Otobius megnini (Dugès, 1883) (spinose ear tick)

General

Otobius megnini is known as the spinose ear tick (Walker *et al.* 2003). There is only one record of *Ot. megnini* in Australia but this is a confirmed record (Mayberry, 2003). It seems that *Ot. megnini* was introduced into Western Australia recently, probably in the ears of horses. The country of origin of these *Ot. megnini* is unknown. Roberts (1970) did not report *Ot. megnini* in Australia. *Otobius megnini* is one of only two species in the genus *Otobius*; *Ot. megnini* is the type species of the genus *Otobius* (Guglielmone *et al.* 2010).

Differential diagnosis

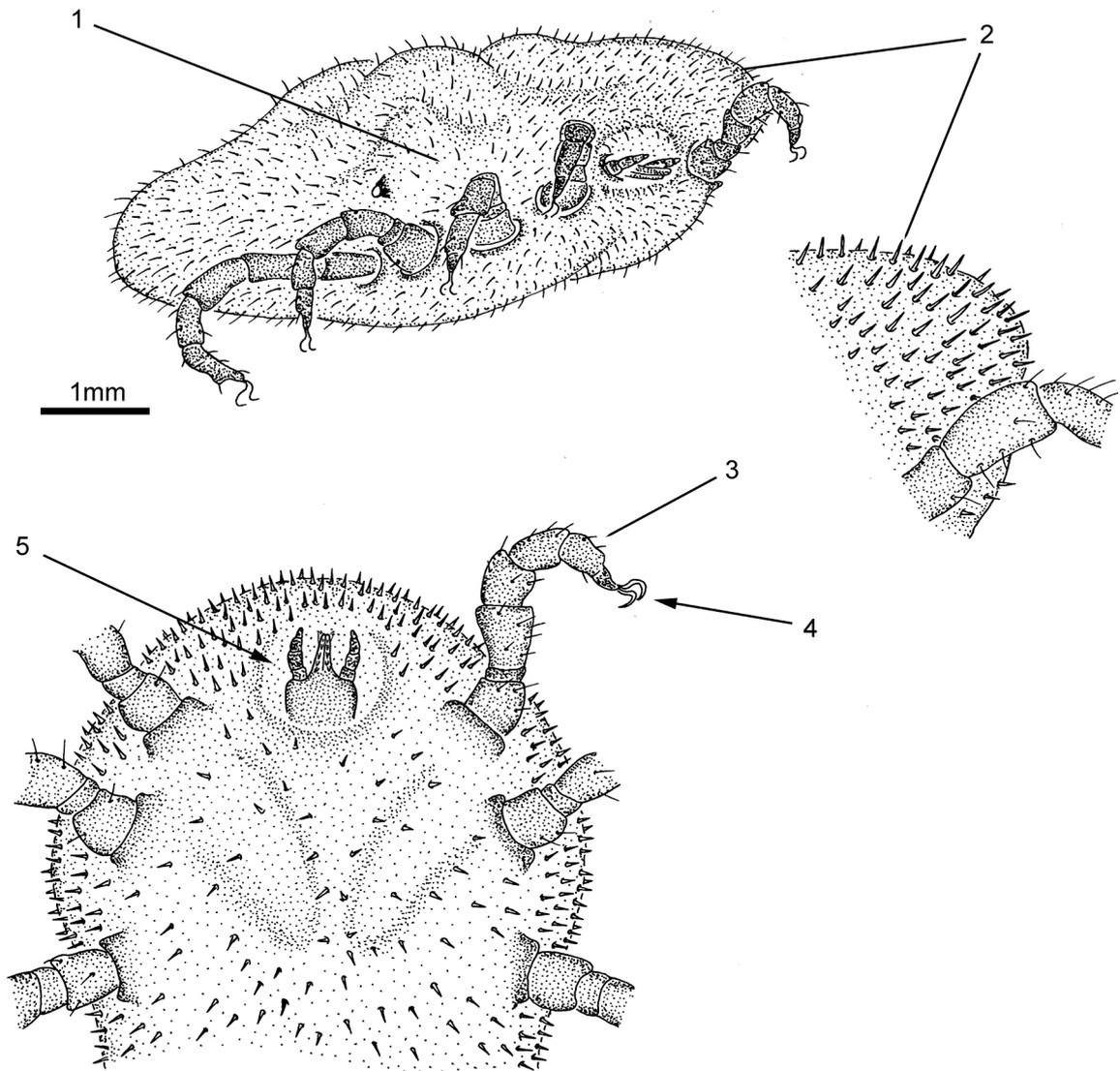
See Figs 10C and 27. *Otobius megnini* is easy to distinguish from all other species of ticks because of its unusual feeding behaviour: only the larvae and nymphs feed on hosts and the adults do not occur on hosts. The larvae have six unusually long legs and mouthparts that protrude anteriorly. As the larvae engorge they become shaped like a violin. The larvae are white or pink in colour. The nymphs are diamond-shaped and then become violin-shaped, similar to the adults. In the nymphs, the mouthparts are ventral and there is no genital aperture. The nymphs are covered with short, rigid setae in the form of spines, from which the tick derives its common name. These setae are less rigid posteriorly than they are anteriorly. The adults are dark grey in colour. Numerous small pits are present on the integument of the adult. The lateral margin of the body is broad, without a definite lateral suture. The adult's mouthparts are rudimentary and situated on the ventral surface of the body. *Otobius megnini* is eyeless.

Hosts

Bos taurus and *B. indicus* (domestic cattle, introduced), *Ovis aries* (sheep, introduced), *Capra hircus* (domestic goats, introduced), *Equus caballus* (horse, introduced), *Equus asinus* (donkey, introduced) and mules (introduced) are the preferred domestic hosts of *Ot. megnini* but this tick may also infest dogs and cats (introduced). Sometimes humans who are exposed through close contact with horses become infested, with painful results. The parasitic larvae and nymphs feed deep in the external ear canal of their hosts. Adult *Ot. megnini* do not feed. In Australia, *Ot. megnini* has been recorded only from horses in Western Australia. As the parasitic stages are only found on close inspection, its presence may remain undetected for a long period.

Life-cycle and seasonality

Otobius megnini is a one-host tick which is domestic, being centred on cattle yards (kraals) and horse stables. The female tick lays 300 to 1,500 eggs, in batches, over a period of months. These eggs are laid in cracks and crevices in yards, pens and stables. The eggs hatch in about three weeks giving rise to six-legged larvae. The larval stage lasts one to two weeks; the larvae moult in the ear canal and give rise to the nymph which is eight-legged and also attaches in the ear. Remarkably, the number of nymphal stages is uncertain but moulting and attachment of nymphs occurs in the ear canal. The nymphs remain in the ear for three to six months and then the nymphs engorge, detach from the ears of their hosts and drop to the ground. The nymphs moult to adults which then mate off the host. Infestations occur throughout the year; the life-cycle takes six months to two years to complete. Feeding behaviour is of the monotropic type. The adults are non-parasitic; they spend their lives in cracks and crevices in ground.



Note: for this one species of the *Otobius* genus in this guide the following features are not part of a comparative character matrix.

- 1 Body is not distinctly divided into dorsal and ventral surfaces.
- 2 Body is covered densely with short thick setae, in form of spines.
- 3 Tarsi of all legs are without dorsal humps.
- 4 Pulvilli are absent.
- 5 Mouthparts are not surrounded by camerostome cheeks.
- * Discs are absent (not labelled).

Note: there is no genital aperture in this figure of a nymph.

FIGURE 27. *Otobius megnini* (spinose ear tick), nymph, lateral and ventral.

Disease

Otobius megnini is not known to transmit any pathogens to its hosts, but ticks feeding in the ear canal and the spines on their bodies may cause considerable irritation. Indeed, infested animals may lose their appetite (= anorexia) and thus lose condition (= cachexia). Ears may become inflamed and tissue necrosis or secondary bacterial infections may develop. These infestations may attract flies to lay eggs, which can lead to fly larva that cause cumulative myiasis-damage (sometimes fatal).

Habitat and geographic distribution

Otobius megnini is endemic to North America where its natural host is *Antilocarpa americana*, the pronghorn antelope. *Otobius megnini* has spread from North America to Australia (WA, Mayberry, 2003), South America, southern, eastern and central Africa, Madagascar, India and Turkey.

Genes and genomes

The following nucleotide sequences are available to the public: (i) large subunit mitochondrial rRNA (16S) (474 bp, Black & Piesman 1994; 272 bp Bastos *et al.* 2009); (ii) large subunit nuclear rRNA (28S) (683 bp, Klompen *et al.* 2000; 1376 bp, Burger *et al.* 2014a); (iii) small subunit nuclear rRNA (18S) (1786 bp Black *et al.* 1997; 1655 bp, Burger *et al.* 2014a); (iv) small subunit mitochondrial rRNA (404 bp, Norris *et al.* 1999); (v) and the entire mitochondrial genome (Burger *et al.* 2014a).

Other information

Nava, Mangold, Guglielmo and colleagues have published extensively on the biology of *Ot. megnini*, for example, Nava *et al.* (2008). Burger *et al.* (2014a) proposed, from entire mitochondrial genome DNA sequences, that *Ot. megnini* is the phylogenetic sister-group to the rest of the subfamily Ornithodorinae (*Ornithodoros sensu stricto* plus the so-called Neotropical Ornithodorinae). Keirans & Pound (2003) is a very useful annotated bibliography of *Ot. megnini* from 1883 to 2000.

Amblyomma triguttatum Koch, 1844 (ornate kangaroo tick)

General.

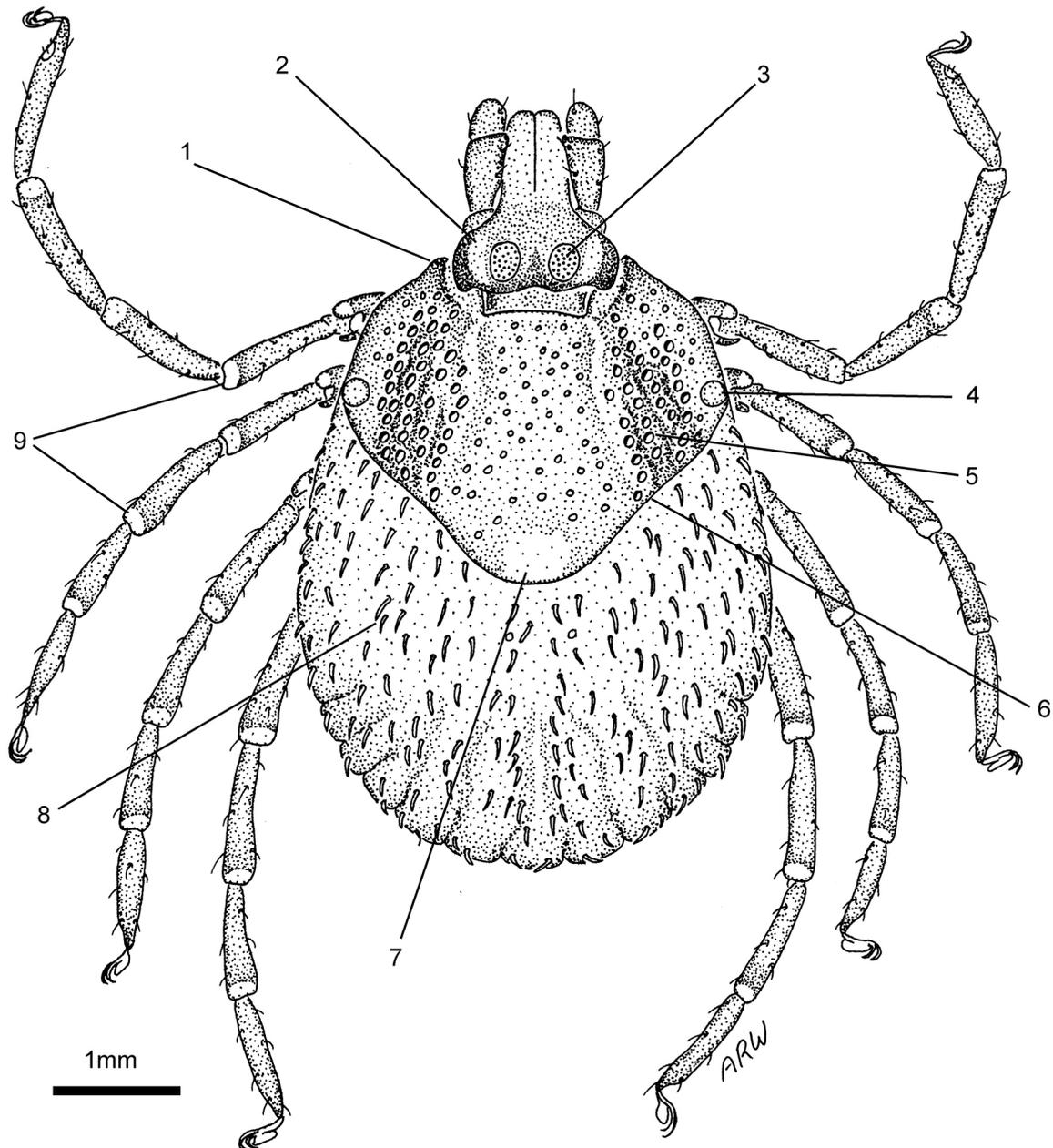
Amblyomma triguttatum is known as the ornate kangaroo tick. This tick has four subspecies: *A. t. triguttatum*, *A. t. queenslandicus*, *A. t. ornatissimum* and *A. t. rosei*.

Differential diagnosis.

There are 18 valid species of *Amblyomma* in Australia. Only one of these species, *A. triguttatum*, is regularly found on domestic animals, mainly cattle. *Amblyomma triguttatum*, however, has been taken from horses and humans (Roberts 1953, 1970). The morphology of *A. triguttatum*, particularly the males, is unique among the known Australian species of *Amblyomma* and indeed among the known *Amblyomma* species of the world (Roberts 1962).

Females. See Figs 11, 28 and 29. The scapulae of the scutum are blunt. The dorsal surface of the basis capituli has a pair of large porose areas and there is a broad shallow groove between each porose area and the lateral margin of the basis capituli. The scutum is densely covered with punctations that are large and deep. The scutum is ornate, usually with a single patch of yellow enamel at the posterior of the scutum but two other small patches may be present. The lateral posterior margins of the scutum are slightly concave. The numerous setae on the alloscutum are flat, long and white. Coxa 1 has two equal and short spurs whilst coxae 2 to 4 have a single short spur each. The spiracle plates are triangular. The hypostome has 4 + 4 columns of teeth.

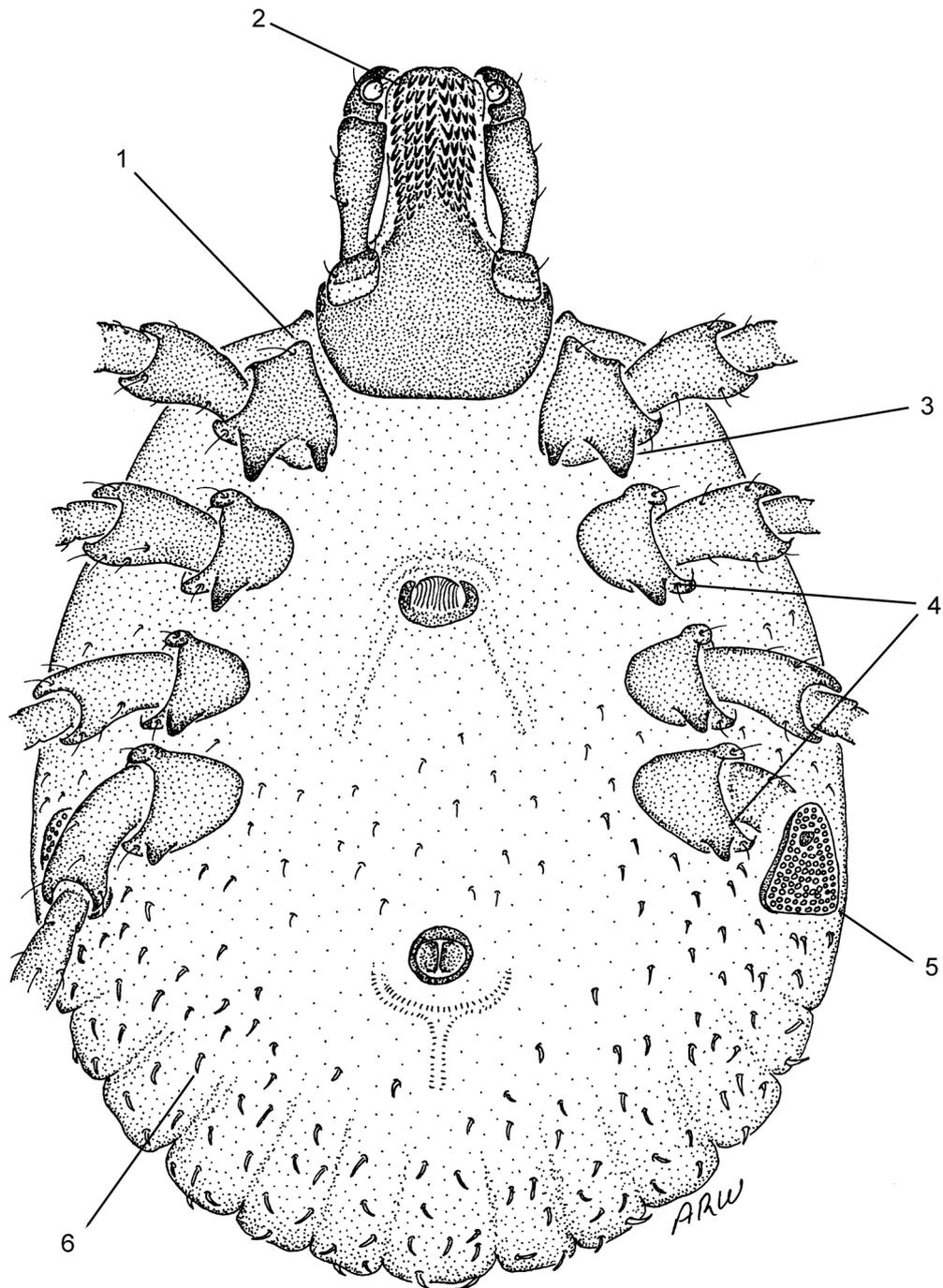
Males. See Figs 11 and 30. The scutum is widely and densely covered with punctations that are large and deep. The scutum is ornate, bearing two small patches of pale yellow enamel in the middle of the scutum and a patch on each festoon. There is a single, shallow, marginal groove (also called a lateral groove) continuous around the posterior of the scutum. This marginal groove consists of a broad line of punctations and fine ridges or striations. Coxa 1 has two equal and short spurs whilst coxae 2 to 4 have a single short spur each. The spiracle plates are comma shaped. The hypostome has 4 + 4 columns of teeth.



Note: for this one species of the *Amblyomma* genus in this guide the following features are not part of a comparative character matrix.

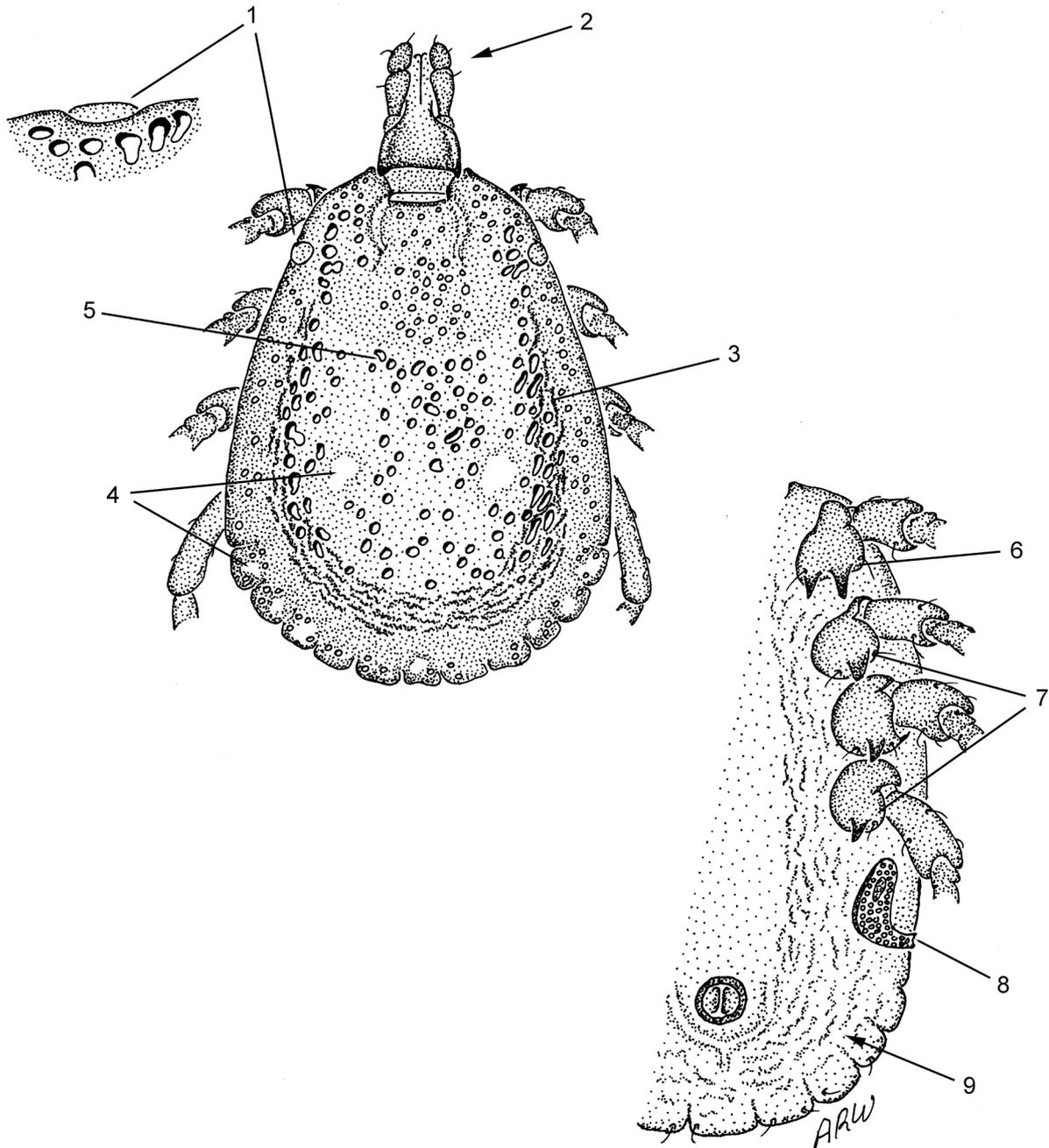
- | | |
|---|---|
| <p>1 Scapulae are blunt.</p> <p>2 Basis has a shallow groove between porose areas and the lateral margins.</p> <p>3 Porose areas are large and separated by same width as a porose area.</p> <p>4 Eyes are flat and touch edge of the scutum.</p> <p>5 Punctations on scutum are large and densely distributed.</p> | <p>6 Posterior margin of scutum is slightly concave.</p> <p>7 Enamel is present as a pale patch at posterior margin of scutum.</p> <p>8 Setae on alloscutum are flat, long and white.</p> <p>9 Legs have narrow pale rings.</p> |
|---|---|

FIGURE 28. *Amblyomma triguttatum* (ornate kangaroo tick), female, dorsal.



- 1 Coxae 1 anterior projection is small and not visible dorsally.
- 2 Columns of teeth on hypostome are 4 + 4.
- 3 Coxa 1 has distinct internal and external spurs.
- 4 Coxa 2 to 4 have distinct external spurs only.
- 5 Spiracle plate shape is triangular.
- 6 Setae on alloscutum are flat, long and white.

FIGURE 29. *Amblyomma triguttatum* (ornate kangaroo tick), female, ventral.



- 1 Eyes are flat and touch edge of scutum.
- 2 (As in female, columns of teeth are 4 + 4).
- 3 Marginal groove is wide, shallow, and marked mainly by fine ridges in the texture of the scutum.
- 4 Enamel is present as two patches on the scutum and patches on some festoons.
- 5 Punctations on scutum are large and deep and densely distributed.

- 6 Coxae 1 have distinct internal and external spurs.
- 7 Coxae 2 to 4 have a distinct external spur.
- 8 Spiracle plate shape is a comma.
- 9 Ventral plaques are absent.

FIGURE 30. *Amblyomma triguttatum* (ornate kangaroo tick), male, dorsal and ventral.

Hosts

Amblyomma triguttatum is a primarily a tick of kangaroos, that is, the large species of the genus *Macropus*. *Amblyomma triguttatum* is also common on wild (feral) pigs in Australia. Indeed, Guglielmone (1990) found that 53 of 146 (36 %) feral pigs in southeast Queensland were infested with *A. triguttatum* and only *A. triguttatum* (314 ticks), except for two individuals of a *Haemaphysalis* species. All 314 of the ticks were nymphs. No larvae or adult ticks were found on these pigs. The vast majority of these nymphs were in the ears of the pigs. Of a sample of 88 grey kangaroos, *Macropus giganteus*, in southeast Queensland, 84% of the kangaroos were infested with larvae, nymphs or adults of *A. triguttatum*, of 1,024 ticks in total. No other species of ticks were found on these kangaroos (Guglielmone 1990). Ninety seven percent of the ticks were in the ears of these kangaroos. McCarthy (1960) also found that *A. triguttatum* prefers to attach in the ears of kangaroos over other sites on the host.

Host-records from Roberts (1962, 1970): *Macropus giganteus* (eastern grey kangaroo), *M. fuliginosus* (western grey kangaroo), *M. robustus* (common wallaroo), *M. rufus* (red kangaroo), *M. dorsalis* (black-striped wallaby), *Myrmecobius fasciatus* (numbat), *Bos taurus* and *B. indicus* (domestic cattle, introduced), *Equus caballus* (domestic horse, introduced), *Ovis aries* (sheep, introduced), *Canis familiaris* (domestic dog, introduced), *Canis lupus* (dingo), *Homo sapiens* (man) and *Ornithorhynchus anatinus* (platypus).

Host-records since Roberts (1962, 1970) (from five different sources): (i) *Macropus agilis* (agile wallaby, Speare *et al.* 1983); (ii) *Wallabia bicolor* (swamp wallaby, Beveridge *et al.* 1985); (iii) *Onychogalea fraenata* (bridled nailtail wallaby, Turni & Smales, 2001); (iv) *Rattus rattus* (black rat, introduced), *Oryctolagus cuniculus* (rabbit, introduced), *M. fuliginosus* (western grey kangaroo), *M. eugenii* (tammar wallaby), *Felis catus* (domestic cat, introduced) and *Canis familiaris* (domestic dog, introduced) (Waudby *et al.* 2007); and (v) *Macropus giganteus* (eastern grey kangaroo), *M. fuliginosus* (western grey kangaroo), *M. robustus* (common wallaroo), *M. rufus* (red kangaroo), *M. eugenii* (tammar wallaby), *Onychogalea fraenata* (bridled nailtail wallaby), *Bettongia tropica* (northern bettong), *Phascolarctos cinereus* (koala), *Ovis aries* (domestic sheep, introduced) and *Homo sapiens* (man) (Barker & Campelo unpublished data).

Life-cycle and seasonality

Amblyomma triguttatum is a three-host tick (Seddon 1968) as are all of the species of *Amblyomma* that have been studied. Male *A. triguttatum* do not feed (Guglielmone & Moorhouse 1986). When *A. triguttatum* copulates, the male inserts only the cheliceral digits into the female's aperture; this is usual in metastriatan ticks i.e. all of the hard ticks except the *Ixodes* spp. (Guglielmone & Moorhouse 1986). Photoperiod in the laboratory, a surrogate for day length, does not affect the time from when females engorge and detach to when they start to lay eggs (the pre-oviposition period) nor the time to egg-hatch (Guglielmone & Moorhouse, 1986). *Amblyomma t. triguttatum* seems to have only one generation each year (Roberts 1962). In both southern Australia (Yorke Peninsula, SA) and northern Australia (Blackall-Barcaldine, central Queensland), adult *A. t. triguttatum* seem to be most abundant in summer (December to February) (McCarthy 1960, Waudby & Petit 2007). In southern Australia (Yorke Peninsula, SA), larvae were most abundant in March (autumn) whereas nymphs were most abundant in September to November (spring).

Disease

Amblyomma triguttatum is a vector of *Coxiella burnetii*, the aetiological agent of Q fever. *Coxiella burnetii* is an intra-cellular bacterium that has a wide host range and a cosmopolitan distribution. In nature, *C. burnetii* cycles between wild animals and ticks. In Australia, *C. burnetii* infects marsupials, particularly macropodids (kangaroos and their kin), sheep, other eutherian mammals and *A. triguttatum* (Derrick *et al.* 1959; Pope *et al.* 1960; Johnson 1966). Shearers may become infected with *C. burnetii* when *A. triguttatum* is cut by the shearer's blades during shearing (fleece removal) of sheep, also contaminating the sheep's wool and the shearing sheds (Pope *et al.* 1960). Perhaps the faeces of *A. triguttatum* also contaminate the sheep's wool and the shearing sheds. Antibodies of *C. burnetii* have been found in a great variety of macropodids: *Macropus agilis*, *M. antilopinus*, *M. dorsalis*, *M. fuliginosus*, *M. giganteus*, *M. robustus*, *M. rufus*, *Petrogale penicillata* and *Thylogale stigmatica* (kangaroos and their kin); *Trichosurus vulpecula* (the common brushtail possum); and *Isoodon macrourus* (northern brown bandicoot) (Cooper *et al.* 2013, 2012a, 2012b); Potter *et al.* 2011). Other hosts of *C. burnetii* include: *Perameles bougainville* (the western barred bandicoot, Bennett *et al.* 2011), *Canis familiaris* (domestic dogs, Cooper *et al.* 2011), *Felis catus* (feral cats), *Vulpes vulpes* (foxes), *Sus scrofa* (feral pigs) and *Canis lupus* (dingoes, Cooper *et al.*

2012b). Banazis *et al.* (2010) found antibodies to *C. burnetii*, and were able to amplify by quantitative polymerase chain reaction (qPCR) the DNA of *C. burnetii* from *M. fuliginosus*. We note that all of these species of mammals are known to be parasitised by one or more species of hard or soft ticks, which thus might be vectors of *C. burnetii* in Australia. Indeed, *A. triguttatum* is known to parasitise six of the nine macropodid species in which antibodies of *C. burnetii* have been found: *M. agilis*, *M. dorsalis*, *M. robustus*, *M. rufus*, *M. giganteus* and *M. fuliginosus* (*A. triguttatum* may well infect the other six species of macropodids too). Cooper *et al.* (2013) noted that transovarial passage of *C. burnetii* has not been reported in Australian ticks, thus far.

Garner *et al.* (1997) reviewed Q Fever, infection of humans with *C. burnetii*, in Australia for the period 1991–4 and this paper is a good introduction to Q Fever in Australia. Beaman & Hung (1989) described an acute case of Q Fever with pericarditis in a man who had been bitten repeatedly by a tick, possibly *A. triguttatum*.

Pearce & Grove (1987), in an elegant paper, described the local skin reactions of 175 soldiers who were bitten by *A. triguttatum* (we presume, *A. t. triguttatum*, based on the known geographic distribution of *A. t. triguttatum*). Moorhouse (1981) also reported local skin reactions, which he described as allergic dermatitis, in humans bitten by *A. triguttatum*.

Rickettsia gravesii, a spotted fever *Rickettsia*, was discovered in *A. triguttatum* (we presume *A. t. triguttatum* based on the known geographic distribution of *A. t. triguttatum*) in Western Australia in 2006. So far this *Rickettsia*, which was named for the Australian *Rickettsia* biologist Stephen R. Graves, has been found only in *A. triguttatum* taken from feral pigs, *Sus scrofa* (feral pigs, Li *et al.* 2010) and humans (Owen *et al.* (2006b) in Western Australia. Owen *et al.* (2006a) suggested that the geographic distribution of *R. gravesii* may coincide with that of *A. triguttatum* (we presume *A. t. triguttatum*, based on the known geographic distribution of *A. t. triguttatum*).

Habitat and geographic distribution

Amblyomma t. triguttatum is known from Queensland, northern New South Wales and Western Australia; *A. t. queenslandicus* from northern Queensland only; *A. t. ornatissimum* from central Queensland and Burketown near the Gulf of Carpentaria, Queensland; whereas *A. t. rosei* is known only from central Australia—see map in Roberts (1962). *Amblyomma t. triguttatum* was recently found on Yorke Peninsula, South Australia (McDiarmid *et al.* 2000).

Genes and genomes

The following nucleotide sequences are available to the public in GenBank small subunit nuclear rRNA (18S) (Dobson & Barker, 1999), and the entire mitochondrial genome (Shao *et al.* 2005b, NC005963).

Other information

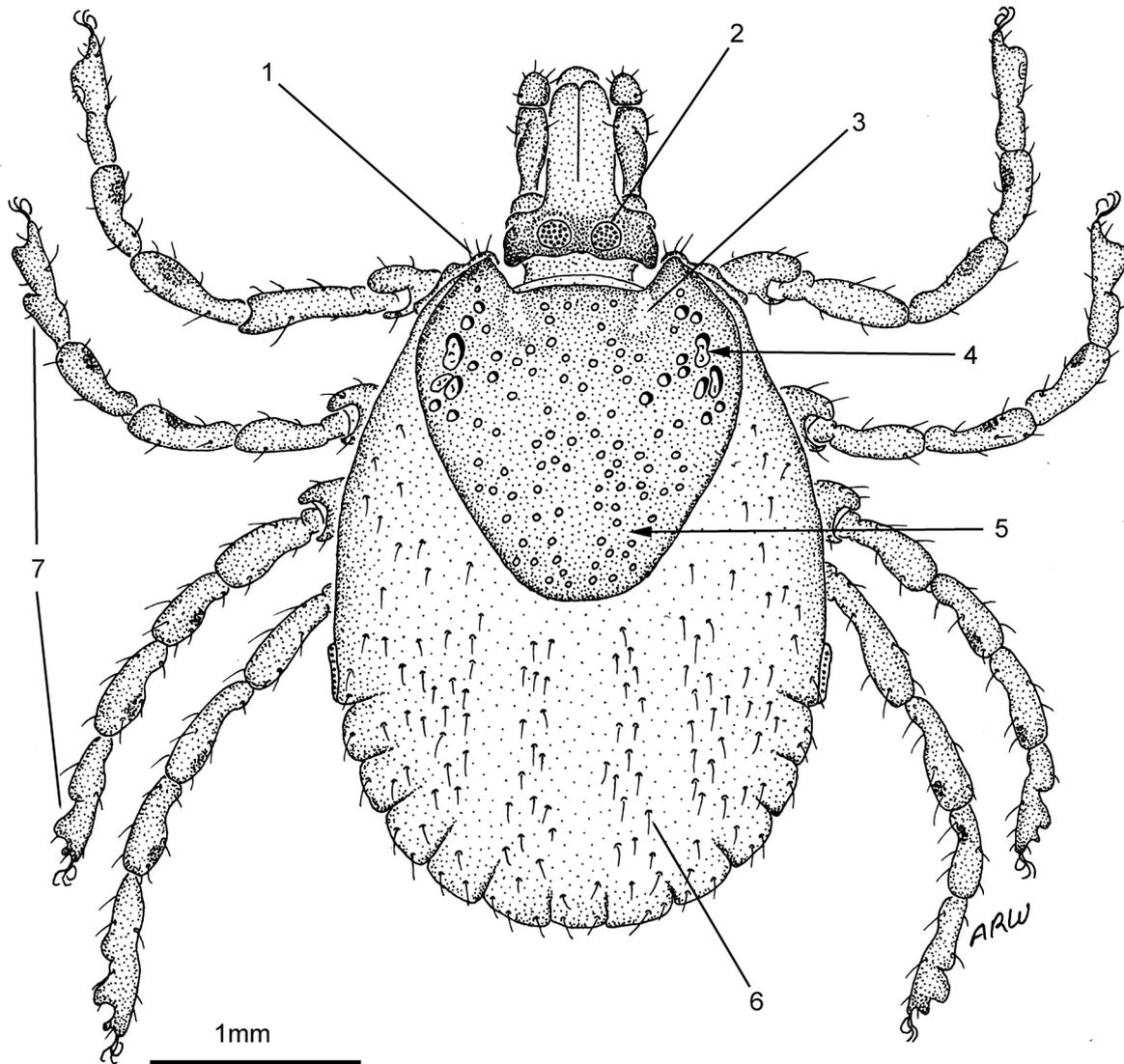
Roberts (1962) described four subspecies of *Amblyomma triguttatum*: *A. t. triguttatum*, *A. t. queenslandicus*, *A. t. ornatissimum* and *A. t. rosei*. Roberts's decision to describe four subspecies was based on the morphology of the capitulum and the scutum of females, the pattern of the colours (ornamentation) of the scuta of females and males, and the observation that each type of tick has a more or less distinct geographic distribution. Less conservative taxonomists would have described a new species rather than a new subspecies. Roberts (1970) emphasized, however, that the subspecies rank for these four types was provisional and that results from cross-breeding experiments or extensive collecting might justify specific status for some or all of the four subspecies. We concur with Roberts (1970) that extensive collecting of ticks, particularly at zones of contact between the different subspecies will resolve the matter of whether all or some of the four subspecies of *A. triguttatum* are species. If some or all of the subspecies of *A. triguttatum* maintain their morphological identity when living together on the same host individual (e.g. kangaroo), then we would consider them to be species rather than subspecies.

Burger *et al.* (2012, 2013) present phylogenetic trees that contain *A. triguttatum* and a dozen or so other species of *Amblyomma*. Waudby *et al.* (2008) studied, by postal survey, the perception and awareness of *A. t. triguttatum* by people in Yorke Peninsula, South Australia. Guglielmone *et al.* (1985) found that attraction by carbon dioxide was superior to dragging cloth through vegetation for catching nymphs and adults of *A. t. triguttatum*. The weight of engorged nymphs is not a reliable indicator of their sex (Guglielmone & Moorhouse 1985). Murrell *et al.* (2003) cultured bacteria from *A. triguttatum* and some other Australian ticks.

***Bothriocroton auruginans* (Schulze, 1936) (wombat tick)**

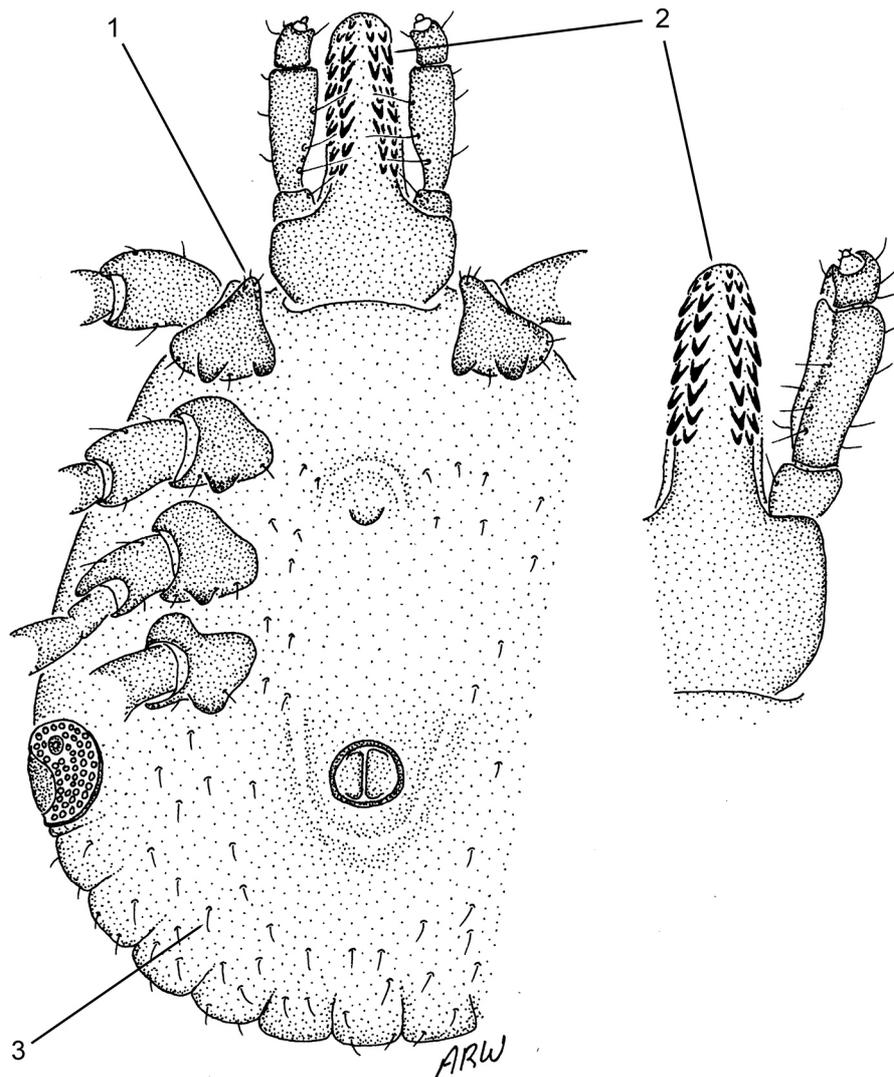
General

Bothriocroton auruginans is known as the wombat tick. Larval and nymphal *B. auruginans* will attach to and feed on domestic dogs that investigate the burrows of wombats.



- 1 Coxa 1 anterior projection is large and visible dorsally.
- 2 Porose areas are narrowly separated.
- 3 Cervical grooves are shallow anteriorly.
- 4 (Eyes are absent in *Bothriocroton* but large punctations can be mistaken for an eye.)
- 5 Enamel on scutum is absent.
- 6 Setae on alloscutum are thin and colourless and sparse.
- 7 Tarsal dorsal humps are large.

FIGURE 31. *Bothriocroton auruginans* (wombat tick), female, dorsal.



1 Coxae 1 anterior projection is large and visible dorsally.

2 Columns of teeth on hypostome are 2 + 2.

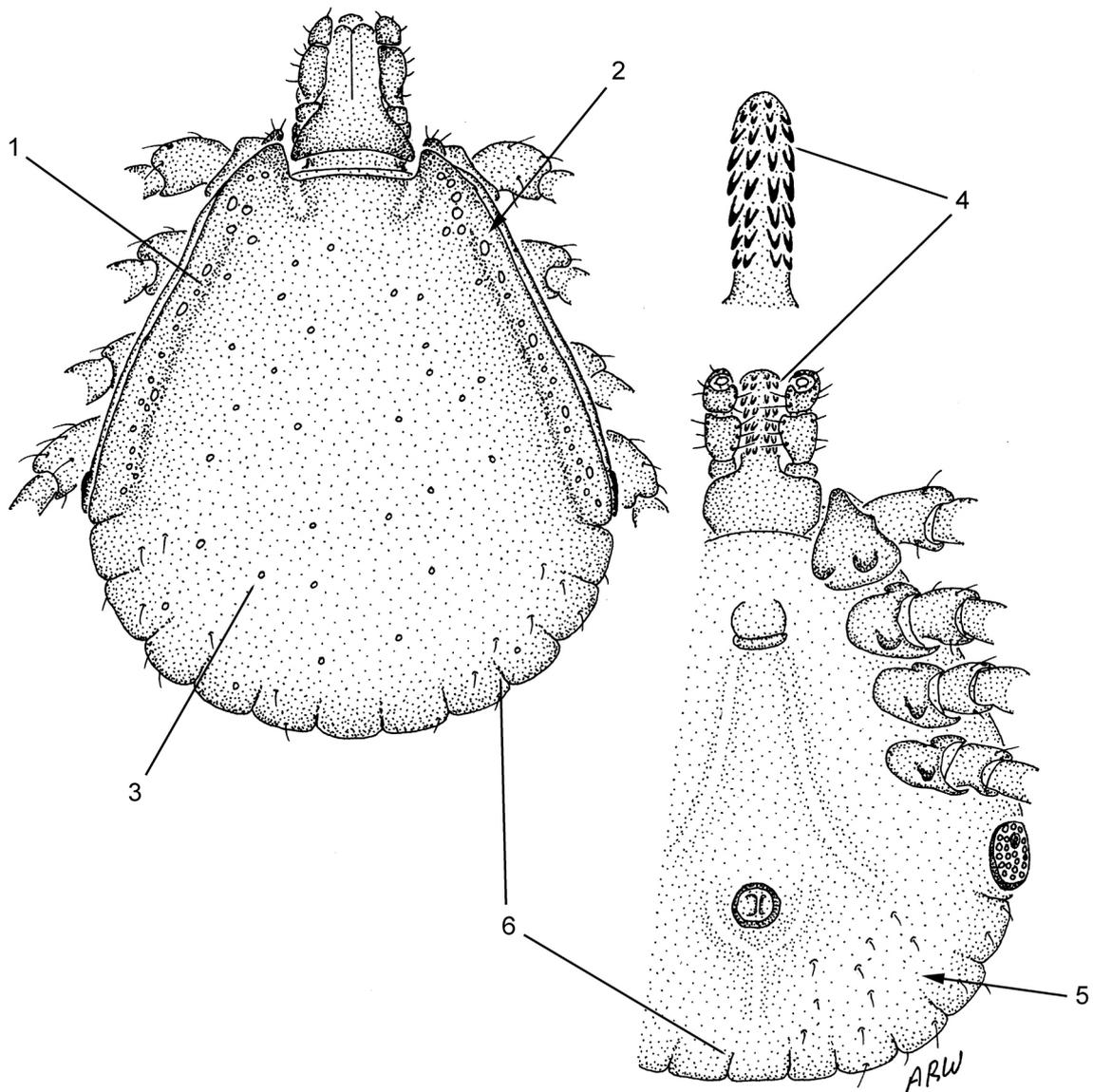
3 Setae on alloscutum are thin and colourless and sparse.

FIGURE 32. *Bothriocroton auruginans* (wombat tick), female, ventral.

Differential diagnosis

See Figs 31 to 33. There are six species of *Bothriocroton* in Australia. *Bothriocroton auruginans* is the only species of *Bothriocroton* to have been recorded from dogs. So if *Bothriocroton* are found on a dog, they are likely to be *B. auruginans*. Furthermore, *B. auruginans* is easily differentiated from *B. hydrosauri*, which may be found on humans. Female *B. auruginans* have a scutum that is without enamel and an alloscutum that has thin colourless setae, also the columns of teeth on the hypostome number 2 + 2. In contrast, *B. hydrosauri* females have a spot of pale enamel at the posterior point of the scutum and an alloscutum conspicuously covered with thick white setae, also the columns of teeth on the hypostome number 4 + 4.

Male *B. auruginans* are without a marginal groove on their conscutum, ventral plaques are absent, and columns of teeth on the hypostome number 2 + 2. Male *B. hydrosauri* have a distinct marginal groove on the conscutum, ventral plaques are present, and columns of teeth on the hypostome number 3 + 3.



- 1 Marginal groove is formed by a line of punctations continuing to first festoon only.
- 2 (Eyes are absent in *Bothriocroton* but large punctations can be mistaken for an eye.)
- 3 Punctations on conscutum are indistinct and sparse.
- 4 Columns of teeth on hypostome are 2 + 2.
- 5 Ventral plaques are absent.
- 6 Festoon grooves are narrow.

FIGURE 33. *Bothriocroton auruginans* (wombat tick), male, dorsal and ventral.

Hosts

Host records from Roberts (1970): *Lasiorhinus latifrons* (hairy-nosed wombat); *Vombatus ursinus* (common wombat), and *Canis familiaris* (domestic dog, larvae and nymphs only). The records of *L. latifrons* are dubious, and thus need to be confirmed with new collections if that is possible, since there was much confusion about the identification of *L. latifrons* and *V. ursinus* in the 1960's when Roberts did his work.

Host records since Roberts (1970): larval *B. auruginans* from *Canis familiaris* (domestic dog, introduced) from Benalla, Dargo, Gippsland, Victoria, and Melbourne, Victoria (I. Beveridge unpublished data). Nymphal *B. auruginans* from *Canis familiaris* (domestic dog, introduced) from Burrawang, New South Wales; *Vombatus ursinus* (common wombat) from Morwell, east Gippsland, Victoria (Smales 1987); and *V. ursinus* (common wombat, Barker & Campelo unpublished data).

Life-cycle and seasonality

Nothing is known about the life-cycle or seasonality of *B. auruginans*. The life-cycle is, however, probably a three-host life-cycle since the other species of *Bothriocroton* that has been studied, *B. hydrosauri*, is a three-host tick (refer to *B. hydrosauri* species account).

Disease

Larval, and to a less extent nymphal, *B. auruginans* may infect *Canis familiaris* (domestic dogs) but ill effects from these infections have not been reported. Vilcins *et al.* (2009a) detected by PCR the DNA of *Coxiella burnetii* and a *Rickettsia* species closely related to *R. massiliae* in *B. auruginans* from Buxton, Victoria. The significance of these findings is not yet clear, but we note that *C. burnetii* and *Rickettsia* species have been found in many Australian mammals and their ticks.

Habitat and geographic distribution

Bothriocroton auruginans is remarkably host specific: adult ticks have only been recorded from wombats so far (Roberts 1970). Given this remarkable host specificity, *B. auruginans* is unlikely to be found far from wombats. Wombats live in wild and rural areas of coastal and sub-coastal southern Australia, mostly from about the New South Wales-Queensland border to the South Australia-Western Australia border and Tasmania (Van Dyke & Strahan 2008). Roberts (1964a) stated that *B. auruginans* existed "throughout the wombat country in New South Wales, extending from Tooloom and Armidale in the north to Bombala in the south. In Victoria specimens have been received from the coastal and sub-coastal areas, including Omeo, Orbost, Healesville, and various localities in the Gippsland district. There are several localities scattered throughout Tasmania and on Flinders Island." For us, this putative geographic distribution needs to be confirmed with specific localities. So far the only confirmed specific localities of *B. auruginans* are: Tooloom and Armidale in New South Wales; Omeo, Orbost and Healsville in Victoria; and Flinders Island, and Tarraleah, Deloraine and Gretna in mainland Tasmania (Roberts 1964b); Benalla, Dargo (Gippsland), and Melbourne in Victoria (Beveridge unpublished data); and Burrawang, New South Wales (Smales 1987). It is notable that *B. auruginans* has not been collected from Queensland, either from *Vombatus ursinus* (common wombat) or from *Lasiorhinus krefti* (northern hairy-nosed wombat).

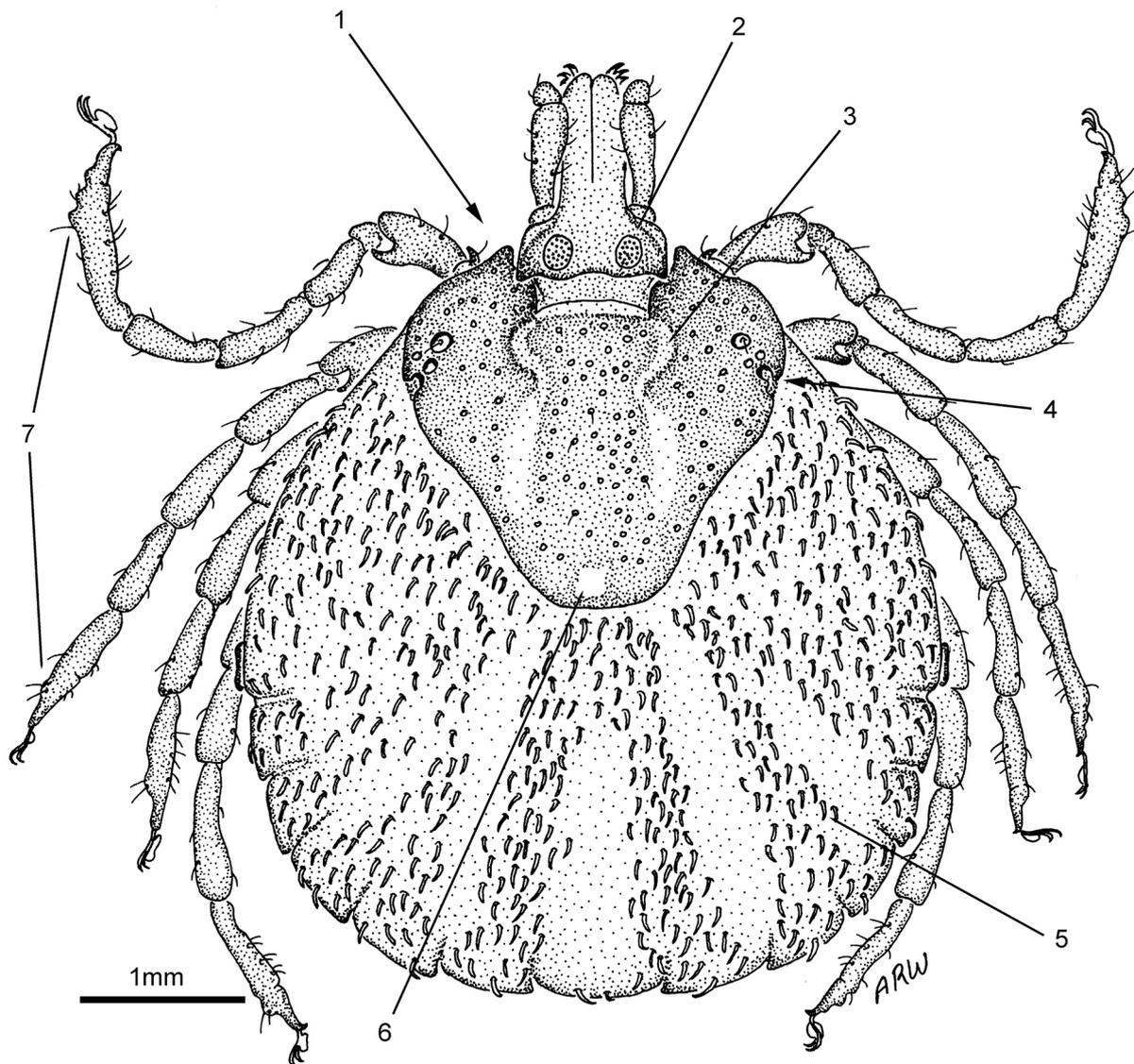
Genes and genomes

Part of only one gene is available to the public, in GenBank: nuclear small subunit nuclear rRNA (18S, 1472 bp) (Klomp *et al.* 2002).

Other information

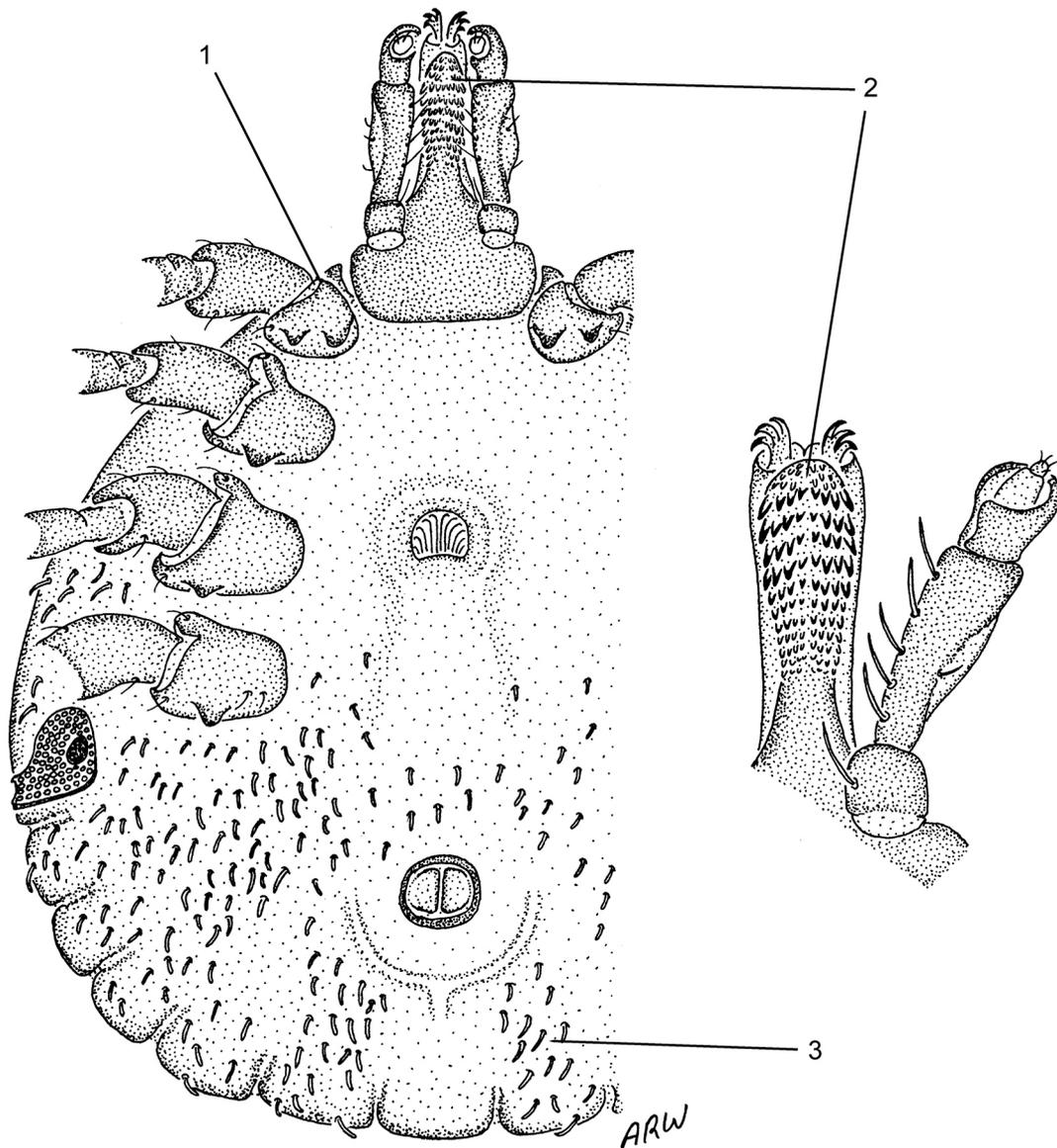
Binnington (1972) discovered putative photoreceptors at the base of all eight legs of adult *B. auruginans* (near coxae I, II, III and IV) in this otherwise eyeless tick.

Bothriocroton hydrosauri (Denny, 1843) (southern reptile tick)



- 1 Coxa 1 anterior projection is small and not visible dorsally.
- 2 Porose areas are widely separated.
- 3 Cervical grooves are deep anteriorly.
- 4 (Eyes are absent in *Bothriocroton* but large punctations can be mistaken for an eye.)
- 5 Setae on alloscutum are broad, white and numerous.
- 6 Enamel on scutum is present, as one small spot at posterior.
- 7 Tarsal dorsal humps are small.

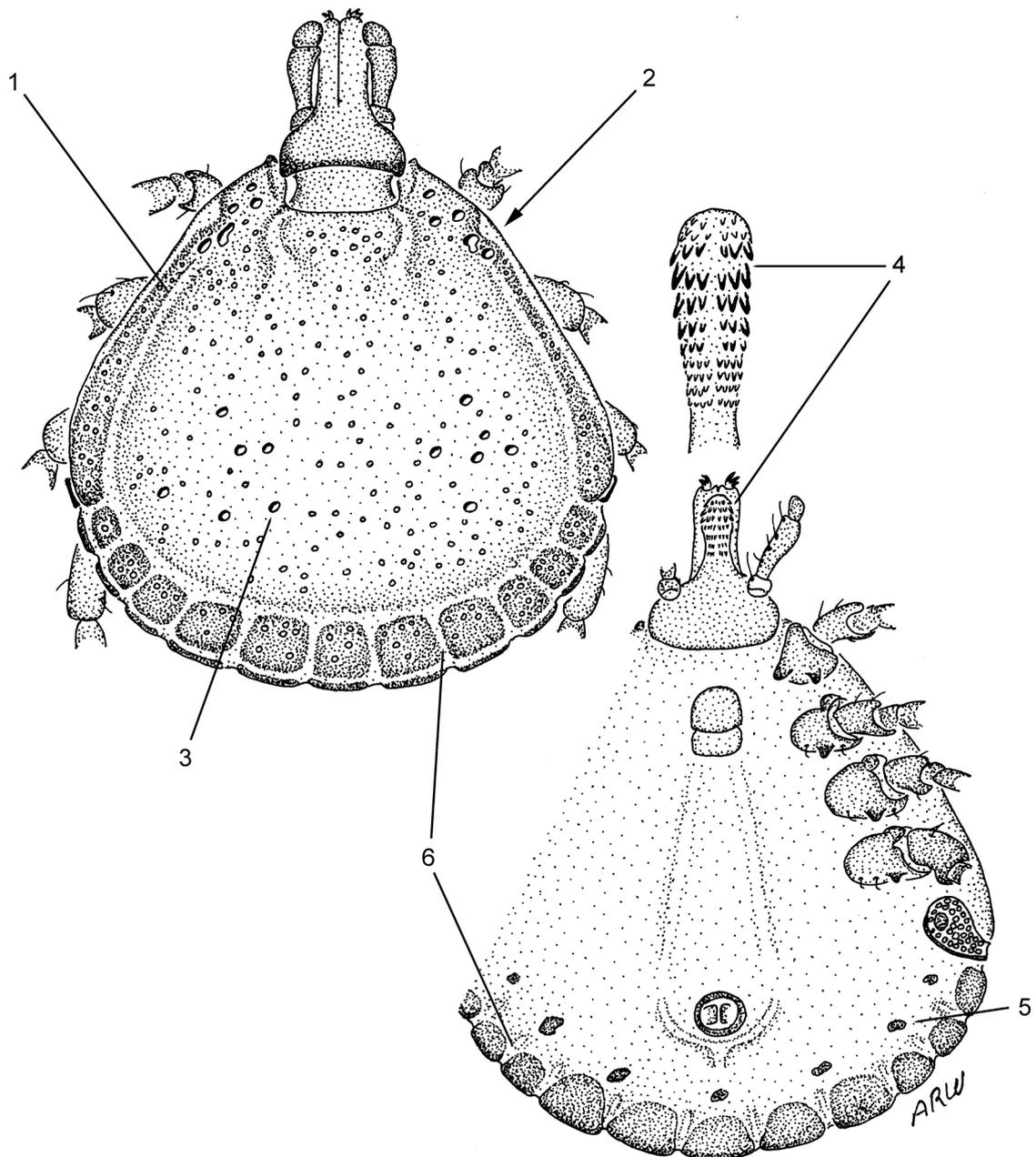
FIGURE 34. *Bothriocroton hydrosauri* (southern reptile tick), female, dorsal.



- 1 Coxa 1 anterior spur is small and not visible dorsally.
- 2 Columns of teeth on hypostome are 4 + 4.
- 3 Setae on alloscutum are broad, white and numerous.

Note: this figure shows the paired sets of cheliceral teeth protruding from the paired cheliceral sheaths.)

FIGURE 35. *Bothriocroton hydrosauri* (southern reptile tick), female, ventral.



- 1 Marginal groove is formed by a deep and smooth groove and continues around anterior edge of all festoons.
- 2 (Eyes are absent in *Bothriocroton* but large punctations can be mistaken for an eye.)
- 3 Punctations on conscutum are distinct and common.
- 4 Columns of teeth on hypostome are 3 + 3 (but 4 + 4 in posterior part of hypostome).
- 5 Ventral plaques are present.
- 6 Festoon grooves are broad.

FIGURE 36. *Bothriocroton hydrosauri* (southern reptile tick), male, dorsal and ventral.



FIGURE 37. Geographic distribution of *Bothriocroton hydrosauri* (southern reptile tick) (redrawn from Bull *et al.* 1981 with two additional localities (Jenolan Caves, NSW, an asterisk; and 38 km south of Gosford, NSW, a cross) from the interpretation of Bull (1965, Table 2) by Andrews *et al.* (2006, p. 37)).

General

Bothriocroton hydrosauri is known as the southern reptile tick. *Bothriocroton hydrosauri* will, however, feed on humans. *Bothriocroton hydrosauri* is notorious on Flinders Island, in South Australia, and perhaps elsewhere in southern Australia, for transmitting to humans *Rickettsia honei*, a spotted fever *Rickettsia*. But *B. hydrosauri* is best known because it is without doubt the most thoroughly studied tick of wildlife in the world. Michael Smyth (1973) discovered the extraordinary geographic and host distribution of *B. hydrosauri*, *Amblyomma limbatum* and *A. albolimbatum*. His PhD student Michael Bull, his students and collaborators subsequently published on this tick and the other ticks it lives with and near, *Amblyomma limbatum* and *A. albolimbatum*, since 1973. There are over 200 papers on almost every aspect of the biology, particularly the ecology, of *B. hydrosauri*. Only some ticks of livestock in other parts of the world have been studied more than *B. hydrosauri*. *Bothriocroton hydrosauri* is a jewel in the crown of Australian tick biology. Most of the papers published on *B. hydrosauri* concern this tick and its reptile host *Tiliqua rugosa*, the sleepy lizard, although *B. hydrosauri* is, as its common name suggests, a generalist reptile tick (Roberts 1953).

Differential diagnosis

See Figs 12, 34 to 36. There are six species of *Bothriocroton* in Australia. *Bothriocroton hydrosauri* is the only

species of *Bothriocroton* known to bite humans. So if *Bothriocroton* are found on humans, they are likely to be *B. hydrosauri*. *Bothriocroton hydrosauri* is easily differentiated from *B. auruginans*, which is the only other species of *Bothriocroton* in this book. Female *Bothriocroton hydrosauri* have a spot of pale enamel at the posterior point of the scutum and an alloscutum conspicuously covered with thick white setae, also the columns of teeth on the hypostome number 4 + 4. Female *B. auruginans* have a scutum that is without enamel and an alloscutum that has thin colourless setae, also the columns of teeth on the hypostome number 2 + 2.

Male *B. hydrosauri* have a distinct marginal groove on the conscutum, ventral plaques are present, and columns of teeth on the hypostome number 3 + 3. Male *B. auruginans* are without a marginal groove on their conscutum, ventral plaques are absent, and columns of teeth on the hypostome number 2 + 2.

Hosts

Bothriocroton hydrosauri was aptly named the southern reptile tick, since it is found on all of the main types of reptiles in southern Australia: lizards, snakes and even a terrestrial turtle. The main host of *B. hydrosauri*, in South Australia at least, is *Tiliqua rugosa* (sleepy lizard). Nonetheless, *B. hydrosauri* will, given the opportunity, attach to feed on humans, cattle and horses.

Host records from Roberts (1970): *Chelodina longicollis* (snake-necked turtle), *Pogona barbata* (common bearded dragon), *Rankinia diemensis* (mountain heath dragon), *Tiliqua scincoides* (common blue-tongue lizard), *T. rugosa* (sleepy lizard), *Varanus gouldii* (Gould's goanna), *Pseudonaja textilis* (common brown snake), *Drysdalia coronoides* (white-lipped snake), *Austrelaps superbus* (lowlands copperhead snake), *Notechis scutatus scutatus* (tiger snake) and *N. s. niger* (tiger snake). The records of *Tachyglossus aculeatus* (echidna), *Equus caballus* (domestic horse, introduced) and *Bos taurus* (domestic cattle, introduced) as hosts of *B. hydrosauri* are seem to be attributable to *B. tachyglossi* (Andrews *et al.* 2006).

Additional host records, since Roberts (1970): (i) *Chelodina longicollis* (snake-necked turtle), *Pogona barbata* (common bearded dragon), *Rankinia diemensis* (mountain heath dragon), *Liopholis whitii* (white's skink), *Niveoscincus metallicus* (metallic skink), *Tiliqua nigrolutea* (southern blue-tongue lizard), *T. occipitalis* (western blue-tongue lizard), *T. scincoides* (common blue-tongue lizard), *T. rugosa* (sleepy lizard), *Varanus gouldii* (sand goanna), *Acanthophis antarcticus* (common death adder), *Pseudonaja textilis* (eastern brown snake), *Drysdalia coronoides* (white-lipped snake), *Diporiphora superba* (superb dragon) and *Notechis* sp. (tiger snake) (Smyth 1973); (ii) *T. rugosa* (sleepy lizard) (Bull & King 1981); (iii) *T. rugosa* (Sharrad & King 1981); (iv) *T. nigrolutea* (southern blue-tongue lizard) and *Notechis scutatus ater* (tiger snake) (Bull 1985); (v) *T. rugosa* (sleepy lizard), *T. scincoides* (common blue-tongue lizard), *Notechis scutatus* (tiger snake) and *Pogona vitticeps* (central bearded dragon); (vi) *Tiliqua adelaidensis* (pygmy blue tongue lizard) (Fenner & Bull 2007); and (vi) *Tiliqua rugosa* (sleepy lizard), *T. scincoides* (common blue-tongue lizard), *Notechis scutatus* (tiger snake) (Barker & Campelo unpublished data).

Life-cycle and seasonality

Bothriocroton hydrosauri is a three-host tick (Bull 1978a, 1978b). More is known about the attachment, courtship and copulation of *B. hydrosauri* on *T. rugosa* (sleepy lizard) than for any other reptile tick (Bull 1986; Chilton & Bull 1991, 1993, 1994; Smallridge & Bull 1999; and in particular Andrews & Bull 1980, 1981). *Tiliqua rugosa* (sleepy lizard) is covered in thick overlapping scales; *B. hydrosauri* is a small tick that can easily crawl under the posterior edges of these scales and then attach to the skin under the scales. Male and female ticks prefer to attach to the axillae of the forelegs or in the ears of their host. Male ticks stay attached for many months waiting for new females to attach to the host. Female ticks must take a small meal of blood (referred to as lymph by Smallridge & Bull 1999) before they are sexually attractive to males. Females that have fed then emit a pheromone which causes males to detach from the host and become active on the surface of the host. The females then use the pheromone to lure males to them. Once a male reaches a female, stereotypic courtship occurs (Fig. 38). This courtship has six phases: (1) contact; (2) dorsal mounting; (3) reversal of position; (4) ventral positioning; (5) pre-copulatory orientation; and (6) copulation. It takes the male 1.5 to 2 hours to transfer his spermatophore to the female genital aperture. Then the six phases of courtship are repeated, but in reverse, until the male reaches the dorsal surface of the female; then he moves away. After copulation, females engorge over 8 to 35 days, apparently on blood and then detach to lay their eggs. Female *B. hydrosauri* lay approximately 1,500 eggs over a period as long as 41 days. Larvae attach to their hosts and engorge. Engorged larvae detach from their hosts. Fifteen to 24 days later, the

larvae moult to nymphs which again attach to their hosts and feed, apparently on blood. Twenty to 33 days after the nymphs have fed again the nymphs moult to adults. Adult ticks then climb back onto their hosts and attach under the scales. Larvae, nymphs and adults may be found together on the same individual host (Bull 1987). Unlike *Ixodes holocyclus* (the paralysis tick), the other native tick that has been studied comprehensively, *B. hydrosauri* does not have a marked season of activity. Copulation, however, is mostly in the spring and summer (September to February) when the host, mainly *T. rugosa*, is most active (Bull & Sharrad 1980, Bull & Burzacott 1994). However, there is some copulation in autumn and winter (March to August). Females that mate in autumn and winter delay egg-laying (go into diapause) for up to six months until the following spring (September to November) when conditions in the soil and leaf litter are favourable for the eggs, and newly hatched larvae are more likely to find a host (Chilton *et al.* 2009). Surprisingly, and most intriguingly, the diapause in egg-laying in *B. hydrosauri* does not occur in *Amblyomma limbatum* which infects the same host, *T. rugosa*, in the same part of South Australia (Chilton *et al.* 2009).

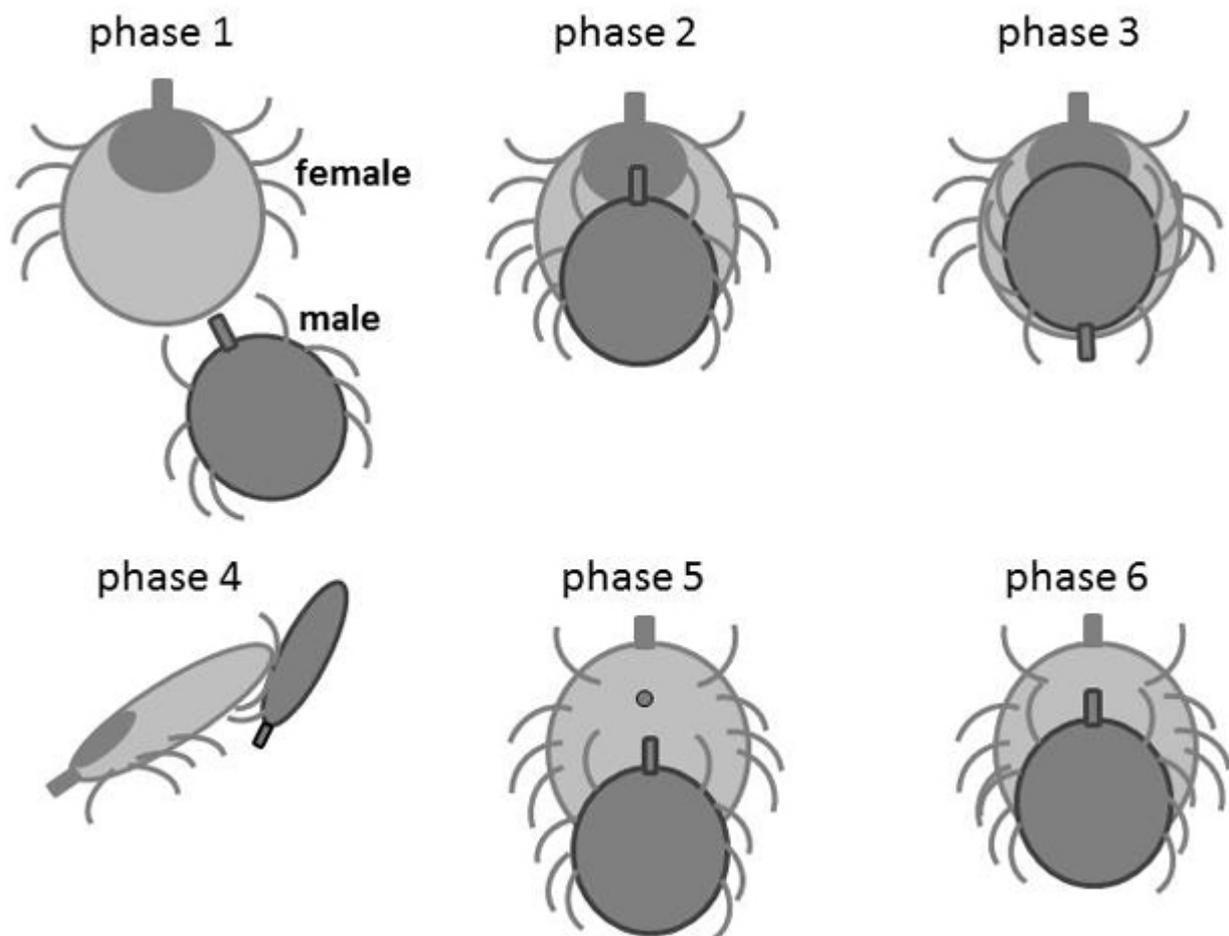


FIGURE 38. Courtship behaviour of *Bothriocroton hydrosauri* (southern reptile tick) (redrawn from Andrews and Bull 1980).

Disease

Bothriocroton hydrosauri is the arthropod-host of *Rickettsia honei* on Flinders Island, Tasmania (Stenos *et al.* 2003) and Tasmania (Whitworth *et al.* 2003). *Rickettsia honei* causes Flinders Island spotted fever in humans (Stenos *et al.* 1998, Unsworth *et al.* 2007b). The common features of Flinders Island spotted fever are head ache, fever, myalgia, cough, arthralgia and maculopapular to purpuric rash with vesiculation; an eschar has been reported in about half of the cases (Graves & Stenos 2003, Parola *et al.* 2005, Murphy *et al.* 2011). Flinders Island spotted fever is typically a relatively mild disease; no deaths have been reported (Graves & Stenos 2009) although a patient in Nepal had severe illness (Murphy *et al.* 2011). *Rickettsia honei* has been isolated from the blood of patients with chronic illness, including fatigue, from Melbourne, Victoria and Adelaide, South Australia, but it is not known whether or not *R. honei* was causally related to the illness (Unsworth *et al.* 2008). Photographs of rashes associated with Flinders Island spotted fever are given in Murphy *et al.* (2011) and Dyer *et al.* (2005).

Curiously, *R. honei* was not detected by PCR nor cell culture in the blood of *Tiliqua nigrolutea* (southern blue-tongue lizard, n=12), *Austrelaps superbus* (copperhead snakes, n=4) nor *Notechis scutatus* (tiger snake, n=3), but 29 of 46 (63%) *B. hydrosauri* from those lizards and snakes were PCR-positive or cell culture-positive for *R. honei* (Stenos *et al.* 2003). *Rickettsia honei* is apparently sustained in populations of *B. hydrosauri* on Flinders Island by trans-ovarial transmission (which is a type of vertical transmission). That is, *R. honei* infects the eggs of *B. hydrosauri in situ* and thus the next generation of *B. hydrosauri* become infected with *R. honei*, without feeding on an infected vertebrate. So, apparently vertebrates are not needed for the survival of *R. honei* on Flinders Island, and perhaps elsewhere. Furthermore, horizontal transmission, that is transmission between the arthropod-host (tick) and the vertebrate-host (lizards and snakes), has not yet been demonstrated experimentally for *R. honei* although there are confirmed cases of infection in people who had been bitten by ticks from Iron Range, Cape York Peninsula, Queensland (*Haemaphysalis novaeguineae*) (Unsworth *et al.* 2007b); and Nepal (species of tick unknown, Murphy *et al.* 2011).

Flinders Island spotted fever is now known from three continents: (i) Australia (Flinders Island, Tasmania, and mainland Tasmania; and South Australia); (ii) Asia (Thailand; Orchid Island, Taiwan; Nepal); and (iii) North America (Texas) (Graves & Stenos 2003; Murphy *et al.* 2011, Dyer *et al.* 2005, Unsworth *et al.* 2005). Confirmed tick-hosts of *R. honei* are: (i) *B. hydrosauri* from Flinders Island and mainland Tasmania, Cooma, New South Wales; Mount Mary, South Australia; (ii) *Haemaphysalis novaeguineae* from Iron Range, Queensland; (iii) *Ixodes granulatus* from *Rattus rattus* (black rat) from Thailand (adult ticks but not yet from larval or nymphal *I. granulatus* (Kollars *et al.* 2001)); and (iv) *Amblyomma cajennense* from cattle from Texas, USA (Stenos *et al.* 2003; Whitworth *et al.* 2003; Graves & Stenos, 2003). Two strains of *R. honei* are known: *R. honei* strain RB from a patient from Flinders Island, Tasmania and *R. honei* strain marmionii from a patient from Iron Range, Cape York Peninsula, Queensland, but doubtless more strains will be recorded in the literature in the future.

Rickettsia honei belongs to the *R. rickettsia* group whereas the other known Australian endemic *Rickettsia*, *R. australis*, belongs to the *R. akari* group (see Parola *et al.* 2005). *Rickettsia honei* is now a model organism for the study of *Rickettsia* species (Frickmann & Dobler 2013a; 2013b; Frickmann *et al.* 2013). The infection of *B. hydrosauri* with *R. coneii* on Flinders Island, Tasmania, is the first record of a *Rickettsia* species in a reptile tick (Stenos *et al.* 2003). The tick-hosts of other *Rickettsia* species are mammal ticks (Stenos *et al.* 2003).

Bothriocroton hydrosauri is the host and vector of at least one other blood parasite, *Hemolivia mariae*. *Hemolivia mariae* is a parasitic protist (Apicomplexa: Coccidia) that lives in the erythrocytes of lizards like *Tiliqua rugosa*. *Bothriocroton hydrosauri* become infected with *H. mariae* when *B. hydrosauri* take a blood meal from an infected lizard. Lizards become infected with *H. mariae* when lizards eat a tick that is infected with *H. mariae* (Smallridge & Bull 1999). Infections of *H. mariae* have a measurable negative affect on the fitness of *Tiliqua rugosa* in semi-arid South Australia. *Hemolivia mariae* infections reduced the activity of individual lizards and thus, the size of their home ranges (territories) (Bouma *et al.* 2007).

The relationships between *B. hydrosauri*, *T. rugosa*, and *H. mariae* are a benchmark for future studies on tick-host-pathogen relationships (Smallridge & Paperna 1997; Smallridge & Bull 1999, 2000, 2001; Godfrey *et al.* 2006; Bouma *et al.* 2007). *Bothriocroton hydrosauri* infections, *per se*, also have a negative effect on the fitness of their lizard hosts. *Bothriocroton hydrosauri* reduces running speed in juvenile *T. rugosa*, and decreases the activity and distance moved each day by these lizards (Main & Bull 2000). Male and female *T. rugosa* typically mate for life; pair-fidelity of 21 years has been recorded (Bull & Burzacott 2006). In some cases, however, female lizards abandon their male partners. Abandoned males have statistically higher ticks loads (*B. hydrosauri*) in their last year with their previous female partner than do males that were able to keep their female partners from one year to the next. Curiously, male lizards did not abandon their female partners when their female partners had high tick loads (Bull & Burzacott 2006).

Habitat and geographic distribution

The geographic distribution of *B. hydrosauri* is by far the best known of a reptile tick anywhere in the world. In South Australia, the boundary of *B. hydrosauri* has been mapped to a scale of metres (Smyth 1973, Bull & King 1981). The most striking feature of the geographic distribution of *B. hydrosauri* on sleepy lizards (*T. rugosa*) is that in South Australia, where it has been studied thoroughly, the distributions of *B. hydrosauri* and the two other species that infect sleepy lizards are parapatric. That is, the geographic distributions of the three species of ticks are mutually exclusive (allopatric), abutting along common boundaries with little (ca. 100 m) or no overlap (Smyth

1973, Bull *et al.* 1981). Moreover those boundaries have not changed for decades (Bull *et al.* 1981, Petney *et al.* 1982). The same abrupt change from *B. hydrosauri* to *Amblyomma albolimbatum* occurs in Western Australia near Albany (Bull & King 1981 cited by Bull 1986 p 394).

There are at least four, probably more, distinct (allopatric) populations of *B. hydrosauri*: (i) southeastern South Australia; (ii) Eyre Peninsula, South Australia; (iii) along the coast from Bremer Bay to Albany, Western Australia; and (iv) Margaret River area, along the coast from Cape Naturaliste to Cape Leeuwin, Western Australia (Smyth 1973, Sharrad & King 1981, Bull 1986; Fig. 37). The simplest explanation for the geographic distribution of *B. hydrosauri* is that present allopatric distributions are relicts of a once much larger, indeed trans-national, geographic distribution (Sharrad & King 1981). One explanation for the apparent reduction in the geographic distribution is competition, and range expansion, by the neighbour of *B. hydrosauri*, *Am. albolimbatum* (Sharrad & King 1981). Bull (1986) discusses the ecological factors that can explain the dramatic tick boundaries of *B. hydrosauri* and its neighbours, *Am. albolimbatum* and *Am. limbatum*.

Genes and genomes

The following nucleotide sequences are available to the public in GenBank: (i) parts of small subunit (12S) and large subunit (16S) mitochondrial rRNA (Norris *et al.* 1999); (ii) large subunit nuclear rRNA (18S) (Klompen *et al.* 2000); (iii) part of *mitochondrial oxidase subunit I* (COI) (Guzinski *et al.* unpublished cited in GenBank); and (iv) nuclear microsatellite markers (Guzinski *et al.* 2008, 2009).

Other information

Curiously, the likely sister-species of *B. hydrosauri* (the southern reptile tick) is *B. tachyglossi* (the northern echidna tick). We can only speculate about the host of the Most Recent Common Ancestor (MRCA) of *B. hydrosauri* and *B. tachyglossi*; this may have been a reptile or an echidna. Binnington (1972) discovered putative photoreceptors at the base of all eight legs of adult *B. hydrosauri* (near coxa I, II, III & IV) in this otherwise eyeless tick. Populations of *B. hydrosauri* may be regulated by density-dependent factors (Tyre *et al.* 2003). GPS loggers attached to individual hosts, genetic data and other methods are currently being used to study the distribution and transmission of *B. hydrosauri* among individual hosts (Guzinski *et al.* 2009, Wohlfiel *et al.* 2013 and references therein).

***Haemaphysalis bancrofti* Nuttall and Warburton, 1915 (wallaby tick)**

General

Haemaphysalis bancrofti is known as the wallaby tick in Australia. *Haemaphysalis bancrofti*, like many species of *Haemaphysalis*, is small and easily missed.

Differential diagnosis

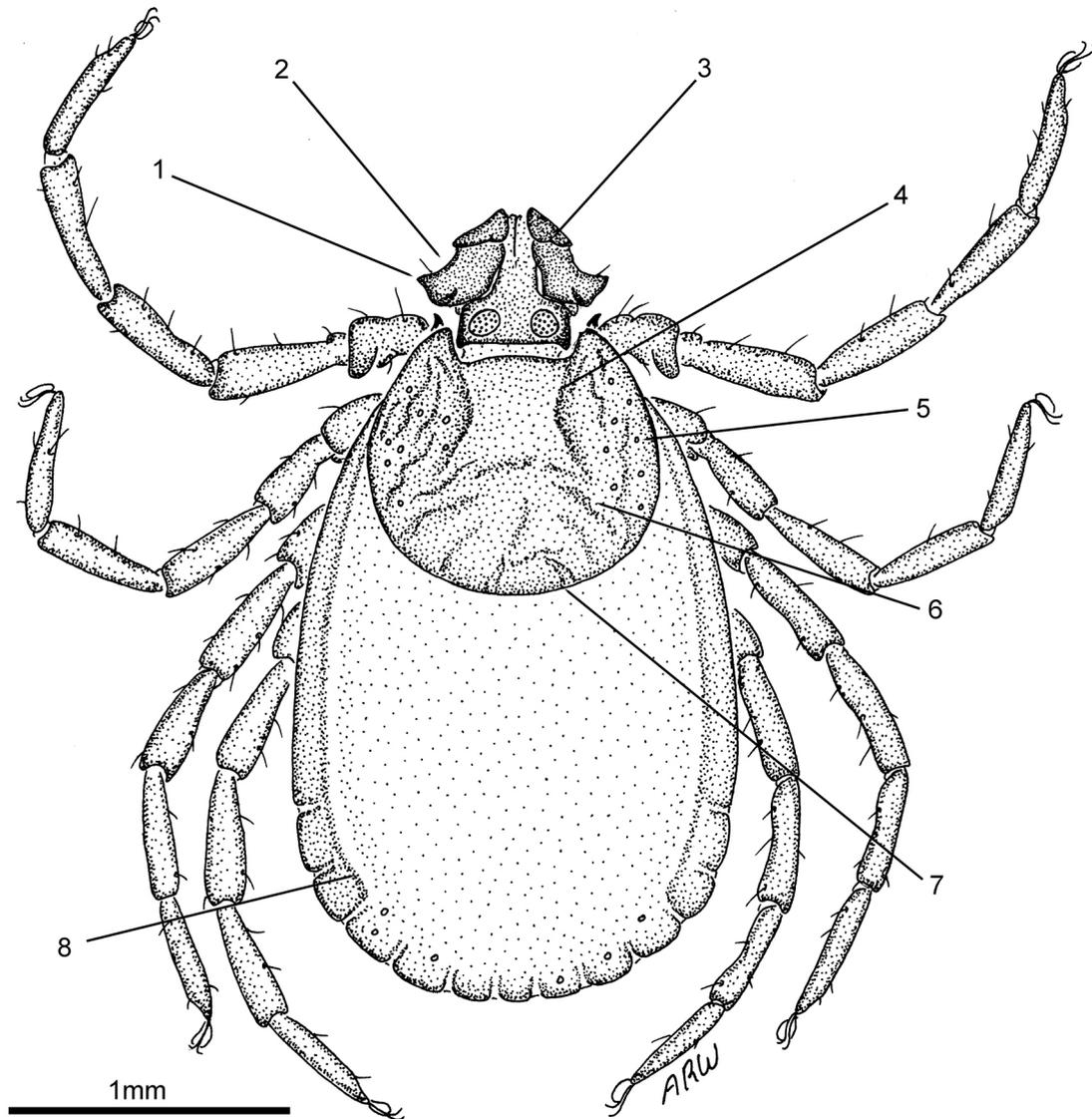
See Figs 14, 39 to 40. *Haemaphysalis* species are small and thus some features may be difficult to see. *Haemaphysalis longicornis* and *H. bancrofti*, the two species of *Haemaphysalis* in this book, however, have distinctive features that differentiate them. Dorsally on *H. bancrofti*, the lateral projection of palp 2 is large; the scutum has a smoothly rounded outline; and there are two festoons enclosed by the lateral groove (these festoons are difficult to see in engorged ticks). Dorsally on *H. longicornis*, the lateral projection of palp article 2 is small; the scutum has a slightly angular outline; and there is only one festoon enclosed by the lateral groove. Ventrally, the most distinctive difference is the hypostome of *H. bancrofti* with 4 + 4 columns of teeth compared to 5 + 5 on *H. longicornis*. Male *H. bancrofti* have one festoon enclosed by the lateral groove compared to none in *H. longicornis*.

Hosts

Haemaphysalis bancrofti is primarily a parasite of the Macropodoidea (wallabies, kangaroos and their kin), but this tick is well adapted to cattle (Roberts 1963, 1970; Barker unpublished data).

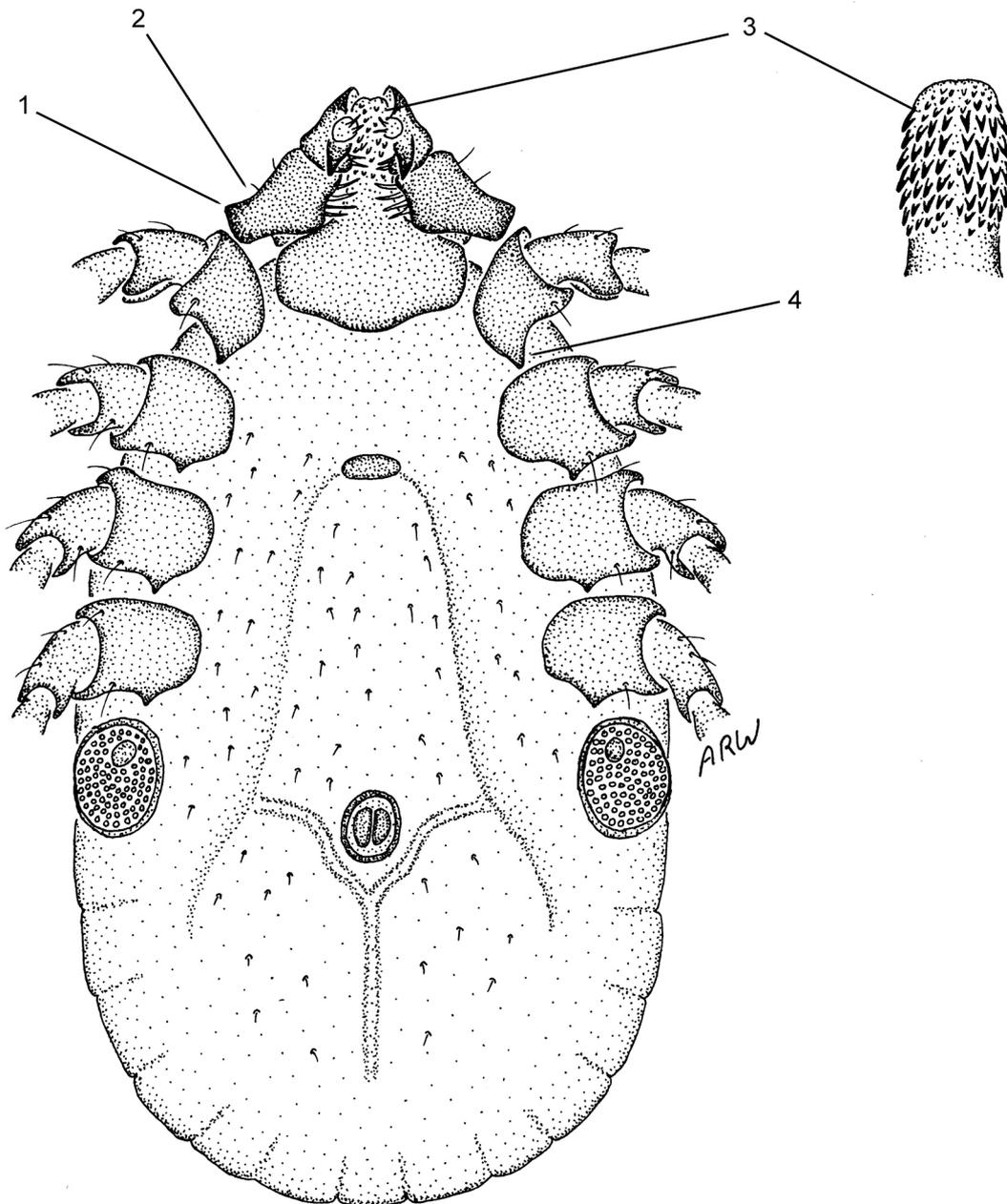
Host-records from Roberts (1963, 1970): *Isodon macrourus* (northern brown bandicoot), *Perameles nasuta* (long-nosed bandicoot), *Macrotis lagotis* (bilby), *Trichosurus vulpecula* (common brushtail possum), *Wallabia bicolor* (swamp wallaby), *Macropus dorsalis* (black-striped wallaby), *M. eugenii* (tammar wallaby), *M. robustus*

(common wallaroo), *M. rufogriseus* (red-necked wallaby), *M. giganteus* (eastern grey kangaroo), *M. parryi* (whiptail wallaby), *Petrogale penicillata* (brush-tailed rock-wallaby), *Potorous tridactylus* (long-nosed potoroo), *Aepyprymnus rufescens* (rufous bettong), *Bettongia penicillata* (woylie), *Thylogale thetis* (red-necked pademelon), *Phascolarctos cinereus* (koala), *Canis familiaris* (domestic dog, introduced), *C. lupus* (dingo), *Equus caballus* (domestic horse, introduced), *Sus scrofa* (pig—feral, introduced), *Ovis aries* (domestic sheep, introduced), *Homo sapiens* (human), a bat *Vespadelus darlingtoni* (= *Esptesicus pumilus*) (tick larvae), and the birds *Centropus phasianus* (pheasant coucal) and *Cracticus tibicen* (Australian magpie).



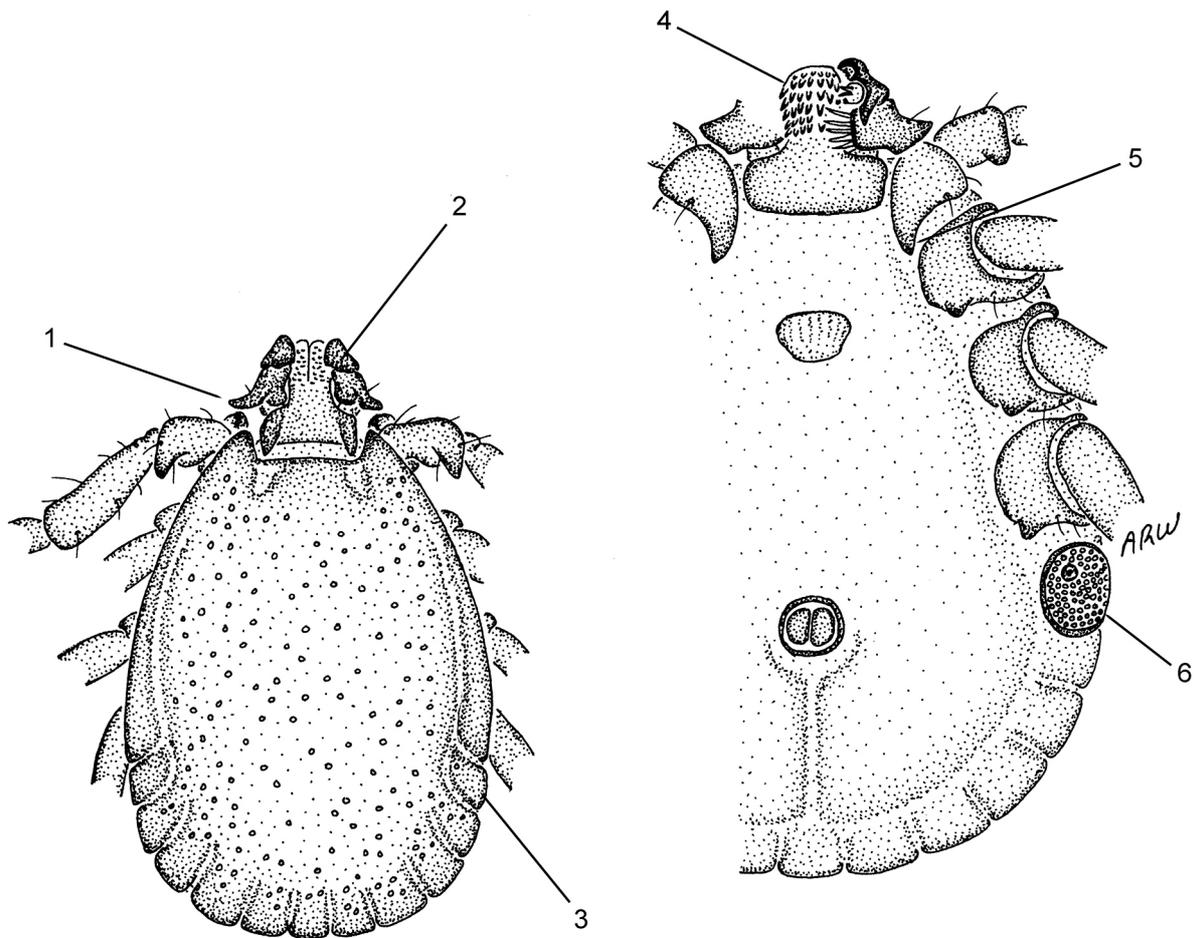
- 1 Palp article 2 lateral projection is large.
- 2 Palp article 2 external margin is distinctly concave.
- 3 Palp article 3 dorsal posterior spur is absent.
- 4 Cervical grooved depth is shallow anteriorly.
- 5 Punctation distribution on scutum is sparse.
- 6 Scutum texture is corrugated.
- 7 Scutum posterior margin is smoothly rounded.
- 8 Festoons enclosed by lateral groove number 2.

FIGURE 39. *Haemaphysalis bancrofti* (wallaby tick), female, dorsal.



- 1 Palp article 2 lateral projection is large.
- 2 Palp article 2 external margin is distinctly concave.
- 3 Columns of teeth on hypostome are 4 + 4.
- 4 Coxa 1 spur profile is bluntly pointed.

FIGURE 40. *Haemaphysalis bancrofti* (wallaby tick), female, ventral.



- 1 Palp article 2 lateral projection is large.
- 2 Palp article 3 dorsal posterior spur is small.
- 3 Fестоons enclosed by lateral groove number 1.
- 4 Columns of teeth on hypostome are 4 + 4.
- 5 Coxa 1 spur profile is bluntly pointed.
- 6 Spiracle plate shape is oval.

FIGURE 41. *Haemaphysalis bancrofti* (wallaby tick), male, dorsal and ventral.

Host-records since Roberts (1970) (from 11 different sources): (i) *Trichosurus vulpecula* (common brush-tailed possum) (Domrow & Smith 1955); (ii) *Macropus agilis* (agile wallaby) (Speare *et al.* 1983); (iii) *Cervus elaphus* (red deer, introduced) (McKenzie *et al.* 1985); (iv) *Wallabia bicolor* (swamp wallaby) (Beveridge *et al.* 1985); (v) *Isodon obesulus* (southern brown bandicoot) and *Rattus rattus* (black rat, introduced) (Heath 1986); (vi) *Dasyurus hallucatus* (northern quoll) (Oakwood & Spratt 2000); (vii) *Onychogalea fraenata* (bridled nailtail wallaby) (Turni & Smales 2001); (viii) *Macropus fuliginosus* (western grey kangaroo) (Oorebeek & Rismiller 2007); (ix) *Petrogale penicillata* (rock-wallaby) (Barnes *et al.* 2010); (x) *Dasyurus maculatus* (spotted-tailed quoll), *Petaurus breviceps* (sugar glider), *Petrogale brachyotis* (short-eared rock-wallaby), *Macropus bernardus* (black wallaroo), *Vombatus ursinus* (common wombat), *Phascolarctos cinereus* (koala) and *Felis catus* (domestic cat, introduced) (Laan *et al.* 2011a); and (xi) *Petrogale assimilis* (allied rock-wallaby), *P. godmani* (Godman's rock-wallaby) and *M. rufogriseus* (red-necked wallaby) (Barker & Campelo unpublished data).

Host-records, Papua New Guinea, from Roberts (1963, 1970): *Sus scrofa* (wild pigs) and *Macropus agilis* (agile wallaby).

Life-cycle and seasonality

Haemaphysalis bancrofti is a three-host tick, as are all of the species of *Haemaphysalis* that have been studied. In southeast Queensland, larvae, nymphs and adults are present all year round (Heath 1986). Thus, it seems that more than one generation is completed each year. In Victoria (Raymond Island), *H. bancrofti* was also present all year round (Laan *et al.* 2011a).

Disease

Haemaphysalis bancrofti is one of the two main vectors of *Theileria orientalis* (= *Theileria buffeli*) in Australia (Stewart *et al.* 1987a, 1989, 1996). The other main vector of *T. orientalis* in Australia is *H. humerosa* (Stewart *et al.* 1987b, 1989, 1996). *Theileria orientalis* is very common in cattle in Queensland, New South Wales & Victoria. For example, Stewart *et al.* (1992) reported that 75% of cattle herds and 41% of the individual cattle they sampled were sero-positive for *T. orientalis*. Infections of cattle with *T. orientalis* are usually benign, but under certain as yet undefined conditions *T. orientalis* may cause serious disease in cattle (Seddon 1966; Izzo *et al.* 2010). Intriguingly, at least four different genotypes of *T. orientalis* occur in Australia: *T. orientalis* (buffeli), *T. orientalis* (ikedai), *T. orientalis* (chitose) and *T. orientalis* (type 4 or type C) (Kamau *et al.* 2011). These genotypes, and/or related genotypes, occur in Japan and Korea (Kamau *et al.* 2011) and likely elsewhere in Asia. The geographic distribution of the genotypes of *T. orientalis* gives weight to the idea that *T. orientalis* was brought to Australia with *H. longicornis* (Seddon, 1966), which is endemic to the Korean Peninsula, Japan and the southern part of the Primorsky Krai Region of Russia and China (Kolonin 2009). Of course there may have been more than one introduction of *H. longicornis* into Australia. The discovery of four genotypes of *T. orientalis* in just 20 cattle (12 cattle from one property in New South Wales and eight cattle from another property in Queensland, Kamau *et al.* 2011), indicates that further sampling will reveal more genotypes of *T. orientalis* in Australia. As argued above, *T. orientalis* was apparently brought to Australia with *H. longicornis*, an Asian tick, yet the main vectors of *T. orientalis* in Australia are *H. bancrofti* and *H. humerosa* which are ticks of native mammals in Australia and Papua New Guinea. It is intriguing that *H. longicornis* is at best an inefficient vector of *T. orientalis* (Stewart *et al.* 1996 and references therein). [Readers should also consult two recent papers that were not reviewed in this present book (Perera *et al.* 2013, 2014)].

Habitat and geographic distribution

Little is known about the preferred habitat of *H. bancrofti* except that most records are from the east-coast of Australia. Laan *et al.* (2011a) proposed that *H. bancrofti* comprises many small disjunct populations. *Haemaphysalis bancrofti* also occurs in Papua New Guinea (Krijgsman & Ponto 1932, Roberts 1963), and there is one record from Java, Indonesia (Kolonin 1978) but this record needs to be confirmed.

Laan *et al.* (2011a) provided a comprehensive list of the published records of *H. bancrofti* (host records and records of free-living ticks), a detailed account of this tick in Victoria (presence and absence of the tick on different species of hosts), and scanning electron micrographs of larvae, nymphs and adults.

Genes and genomes

GenBank does not contain any genes of *H. bancrofti*.

Other information.

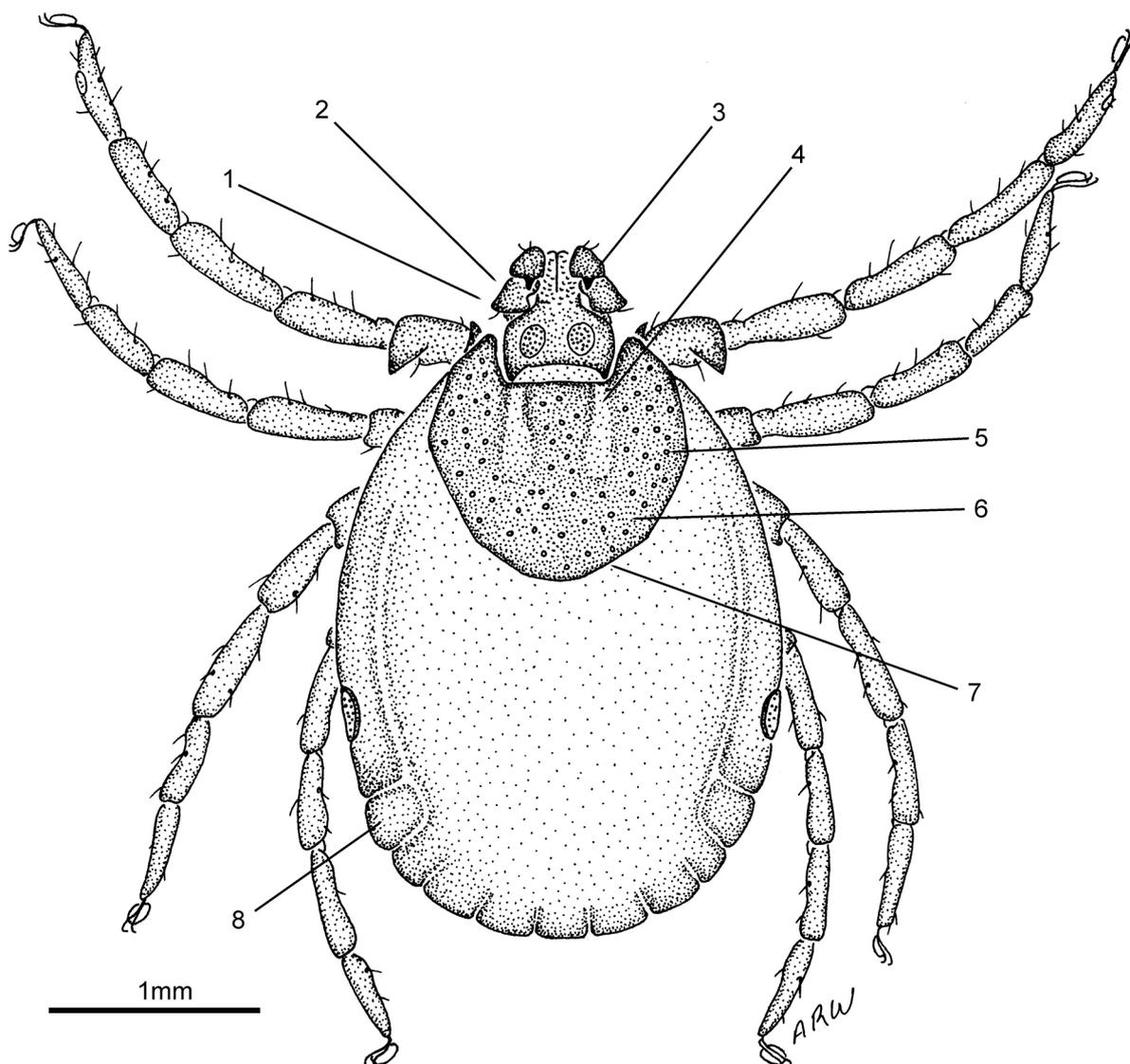
None.

***Haemaphysalis longicornis* Neumann, 1901 (bush tick)**

General.

In Australia, *Haemaphysalis longicornis* is known as the bush tick. Curiously, the type locality of *H. longicornis* is Kempsey in New South Wales but this tick certainly came to Australia from somewhere in Asia in the 19th or 20th

centuries (see Habitat and geographic distribution, below). *Haemaphysalis longicornis* is one of 42 species in the subgenus *Kaiseriana* (Camicas *et al.* 1998).



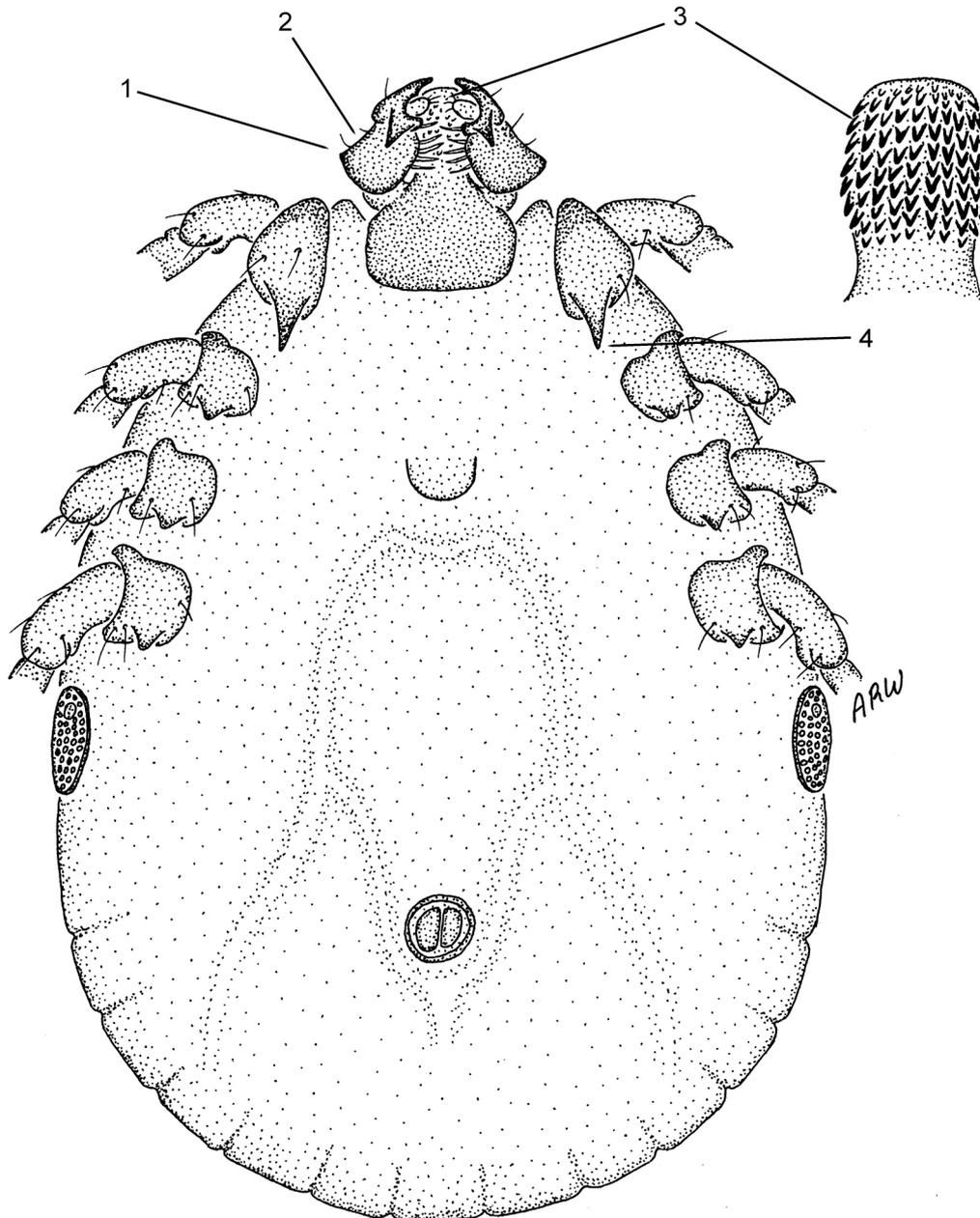
- 1 Palp article 2 lateral projection is small.
- 2 Palp article 2 external margin is slightly concave (best seen from ventral view).
- 3 Palp article 3 dorsal posterior spur is present.
- 4 Cervical grooves are deep anteriorly.
- 5 Punctation distribution on scutum is dense.
- 6 Scutum texture is smooth.
- 7 Scutum posterior margin is slightly angular.
- 8 Festoons enclosed by lateral groove number 1.

FIGURE 42. *Haemaphysalis longicornis* (bush tick), female, dorsal.

Differential diagnosis.

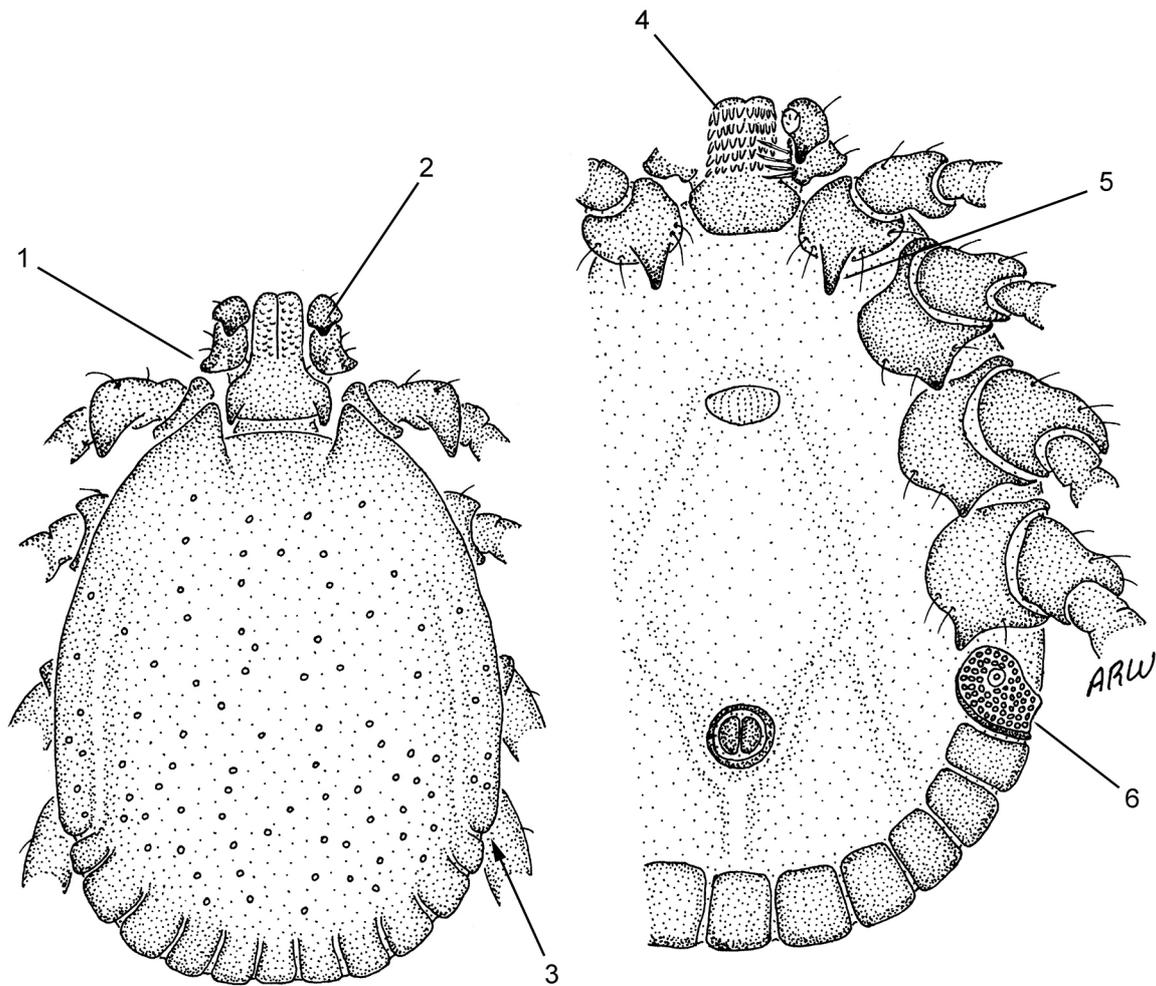
See Figs 42 to 44. *Haemaphysalis* species are small and thus some characters may be difficult to see. Two species of *Haemaphysalis* infect domestic animals in Australia: *H. longicornis* and *H. bancrofti*. Fortunately, *H. longicornis* and *H. bancrofti* have distinctive features that differentiate them. Dorsally on *H. longicornis*, the lateral projection of palp article 2 is relatively small; the scutum has a slightly angular outline; there is only one festoon

enclosed by the lateral groove. Dorsally on *H. bancrofti*, the lateral projection of palp 2 is large; the scutum has a smoothly rounded outline; there are two festoons enclosed by the lateral groove (these festoons are difficult to see in engorged ticks) Ventrally, the most distinctive difference is the hypostome of *H. longicornis* with 5 + 5 columns of teeth, compared to 4 + 4 on *H. bancrofti*. See the account for *H. bancrofti* for a comparison of males, but males of *H. longicornis* are unlikely to be found for comparisons since in Australia the females of this tick reproduce by parthenogenesis, without need for males (Bremner 1959).



- 1 Palp article 2 lateral projection is small.
- 2 Palp article 2 external margin is slightly concave.
- 3 Columns of teeth on hypostome are 5 + 5.
- 4 Coxa 1 spur profile is sharply pointed.

FIGURE 43. *Haemaphysalis longicornis* (bush tick), female, ventral.



- 1 Palp article 2 lateral projection is small.
- 2 Palp article 3 dorsal spur is large.
- 3 Festoons enclosed by lateral groove number 0.
- 4 Columns of teeth on hypostome are 5 + 5.
- 5 Coxa 1 spur profile is sharply pointed.
- 6 Spiracle plate shape is like a comma.

FIGURE 44. *Haemaphysalis longicornis* (bush tick), male, dorsal and ventral.

Hosts

Cattle are the preferred hosts of *H. longicornis* in Australia but sheep and horses may also be heavily infected (Roberts 1970). *Haemaphysalis longicornis* has also been collected from *Homo sapiens* (human), *Felis catus* (domestic cat, introduced), *Canis familiaris* (domestic dog, introduced), *Sus scrofa* (domestic pig, introduced), *Lepus europaeus* (European hare, introduced) and some "marsupials" (Roberts 1970). Larvae have been found on: (i) domestic fowl (*Gallus gallus*), domestic ducks (*Anas platyrhynchos*), turkeys (*Meleagris gallopavo*) and domestic pheasants (*Phasianus colchichus*) (Myers 1924 in Heath *et al.* 1988); (ii) European house sparrows (*Passer domesticus*, introduced) and Australian magpies (*Cracticus tibicen*) (Roberts, 1970); and (iii) the North Island brown kiwi (*Apteryx australis mantelli*) and buff-banded rail (*Rallus philippensis assimilis*) of New Zealand (Heath *et al.* 1988). Nymphs have been taken from Australian magpies (Roberts 1970). Cattle, sheep, pigs, poultry

and dogs are potential and/or actual carriers of *H. longicornis* across seas and oceans. Indeed, a live female *H. longicornis* was shipped to Hawaii on a sheep-dog from Australia that was destined for Texas, USA (Hoogstraal *et al.* 1968).

Life-cycle and seasonality

Haemaphysalis longicornis is a three-host tick as are all of the *Haemaphysalis* species that have been studied. *Haemaphysalis longicornis* in Australia is an obligate parthenogen; that is, the females lay fertile eggs without the service of male ticks (Bremner 1959). Indeed, male *H. longicornis* are rare; Bremner (1959) found one male for every 400 or so females. The following account is that of Arundel and Sutherland (1988) who summarised the work of Roberts (1952) and Wagland *et al.* (1979). Eggs of *H. longicornis* take 37–90 days to hatch. Larvae attach to a host, most larvae engorge in four days, detach and fall to the ground where they moult to nymphs in 19–22 days. The nymphs then attach to a host, engorge for 4–5 days, detach and fall to the ground where they moult to young adults in 23–95 days. Unfed larvae can survive for up to 217 days, unfed nymphs up to 263 days and unfed adult females up to 249 days. Females take 5–6 days to engorge. Other pertinent papers on the life-cycle and seasonality of *H. longicornis* in Australia include: Sutherst and Bourne (1991) on the development, fecundity and behaviour of *H. longicornis* in southeast Queensland and Heath (1979, 1981) on the temperature and humidity preferences of *H. longicornis* in the laboratory.

Disease

Haemaphysalis longicornis was long thought to be a vector, perhaps the main vector, of *Theileria orientalis* in New South Wales. But extensive field and laboratory studies by Stewart and colleagues showed that if *H. longicornis* is indeed a vector of *T. orientalis* in Australia, it is at best an inefficient vector in this country (Stewart *et al.* 1996 and references herein). In New Zealand, however, *H. longicornis* is indeed apparently a vector of *T. orientalis* since *H. longicornis* is the only tick that typically parasitises cattle in New Zealand (James *et al.* 1984). Either *H. longicornis* from New Zealand and *H. longicornis* from Australia differ in vector-competence for *T. orientalis* or the *T. orientalis* populations in New Zealand and Australia differ in their ability to infect this tick. Certainly, there are many different genotypes of *T. orientalis*; Kamau *et al.* (2011) found four different genotypes of *T. orientalis* in cattle from Australia. See the species account of *H. bancrofti* for information on *T. orientalis* in Australia.

Haemaphysalis longicornis is considered to be a vector of viruses, bacteria and other protozoa (e.g. *Babesia gibsoni*) in other parts of the world, particularly in Asia where *H. longicornis* apparently evolved. None of these pathogens have been shown to be transmitted by *H. longicornis* in Australia.

Habitat and geographic distribution

Haemaphysalis longicornis is endemic to the southern part of the Primorsky Krai Region of Russia, Korean Peninsula (North and South Korea), Japan, and China (see Kolonin 2009). Hoogstraal *et al.* (1968) proposed that *H. longicornis* came to Australia from northern Japan in the 19th century and then to New Zealand, New Caledonia and Fiji. *Haemaphysalis longicornis* may also occur in Tonga, New Hebrides and Samoa (Kolonin, 2009, but these records need to be confirmed). It is not known on which host *H. longicornis* reached Australia and the islands listed above (see note above on potential and actual hosts that might carry this tick across oceans). Comparison of mitochondrial *cytochrome c oxidase subunit I (cox1)* sequences of *H. longicornis* from the different islands it inhabits and from its endemic area in Asia, may allow the invasion-history of this tick to be unravelled (Burger *et al.* 2014b serves to highlight the potential power of *cox 1* and other mitochondrial genome sequences for inferring the evolutionary history of ticks).

In eastern Australia, *H. longicornis* occurs in a narrow coastal strip, about 100 km wide, from Gympie, Queensland, in the north to the Wodonga-Tallangatta region in the south (Roberts 1970, Dicker 1978). In Queensland and New South Wales, *H. longicornis* is abundant, in high rainfall areas like Tamborine, Buderim and Maleny in Queensland, and Taree and Wauchope in New South Wales. In Victoria, heavy infestations of cattle, that required treatment, have been recorded at Corryong, but elsewhere in Victoria *H. longicornis* usually occurs on cattle in low numbers. *Haemaphysalis longicornis* seems to thrive only in regions with high summer rainfall and moderate temperatures (Sutherst pers. comm. cited by Beiser & Wroth 1985). In the Taree-Wauchope region of New South Wales, large numbers of adult ticks are found on cattle in spring (September to November) and summer (December to February) (Dicker 1978). Larvae are found on cattle from mid-summer (about January) to mid-

autumn (about April). Nymphs spend the winter (June to August) in the pasture. In Western Australia, *H. longicornis* is restricted to a coastal area between about Walpole and Denmark that is only about 60 km long and 20 km wide (Beiser & Wroth 1985, Beiser 2013 pers. comm.). *Haemaphysalis longicornis* was first reported in Western Australia in 1983 (Beiser & Wroth 1985) although it may have been present in Western Australia before 1983. Heavy infections were reported in 1983: up to 300 ticks on individual cattle, 100 ticks on individual sheep and 100 ticks on dogs on 12 farms (Beiser & Wroth 1985). Intriguingly, heavy infestations have not occurred since about 1983–5 (Beiser 2013 pers. comm.). *Haemaphysalis longicornis* has not spread from the Walpole-Denmark region to other parts of Western Australia. This was the prediction of the CLIMEX computerised simulation in 1983 (Beiser & Wroth 1985, Beiser 2013 pers. comm.).

Genes and genomes

The genes of *H. longicornis* have been studied more than those of most ticks; 669 sequences were in GenBank and thus available to the public at the time of writing, including much of the mitochondrial genome (Murrell *et al.* 2003).

Other information

Many people work on *H. longicornis* in the laboratory and in the field, so there is a very large and growing literature on this species, particularly in China where this tick is widespread. A PubMed search on 14 March 2014 revealed 298 papers on *H. longicornis*. *Haemaphysalis longicornis* has been well studied in New Zealand: Bull (1953), Heath (1978, 1994, 2013), Heath *et al.* (1977, 1987, 1988, 2011), Neilson & Mossman (1982), Hardwick (2010) and McFadden *et al.* (2011). Dicker and Sutherst (1981) reported on experiments in which *Bos taurus* × *B. indicus* cattle carried one quarter of the *H. longicornis* as pure *B. taurus* cattle. Wagland *et al.* (1985) showed that cattle infected with *H. longicornis* were just as susceptible to *Rhipicephalus australis* as cattle that had never been exposed to *H. longicornis* or *R. australis* i.e. there was no evidence of cross-resistance between *H. longicornis* or *R. australis*. Burger *et al.* (2013 and references therein) offer hypotheses about the evolutionary history (phylogeny) of *H. longicornis* and its kin. Murrell *et al.* (2003) cultured bacteria from *H. longicornis* and some other Australian ticks.

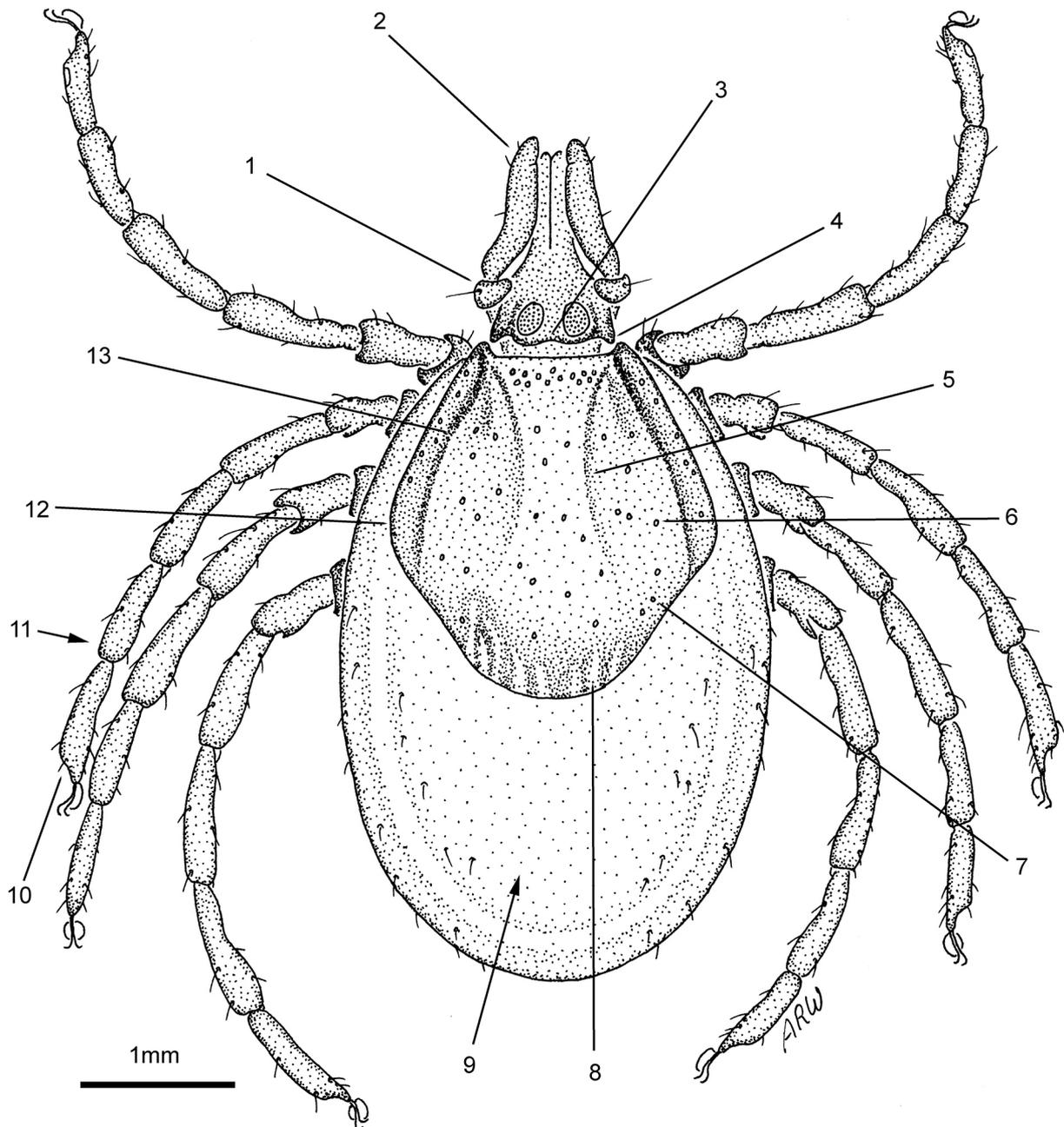
Ixodes cornuatus Roberts, 1960 (southern paralysis tick)

General.

Ixodes cornuatus is known as the Tasmanian paralysis tick (Carne *et al.* 1987) which is a misnomer since this tick also occurs on mainland Australia. Thus, we propose that the common name of *I. cornuatus* be “southern paralysis tick” rather than “Tasmania paralysis tick”. *Ixodes cornuatus* is the only tick that has been associated, clinically, with paralysis in Tasmania, although we note that some of the other six species in the subgenus *Sternalixodes* occur in Tasmania e.g. *I. hirsti*. It has been hypothesised that all seven species of the subgenus *Sternalixodes* may cause paralysis (Kemp 1979) but so far only *I. cornuatus*, *I. holocyclus* and *I. hirsti* have been associated, clinically, with paralysis in their hosts. *Ixodes cornuatus* is one of 28 species in the Australasian lineage of the genus *Ixodes* (Barker & Murrell 2004, 2008) and one of seven species in the subgenus *Sternalixodes*, all of which occur only in Australia, except for *I. confusus* and *I. cordifer* which also occur in Papua New Guinea. *Ixodes cornuatus* is said to be the most common tick on domestic animals in Tasmania (Arundel & Sutherland 1988).

Differential diagnosis.

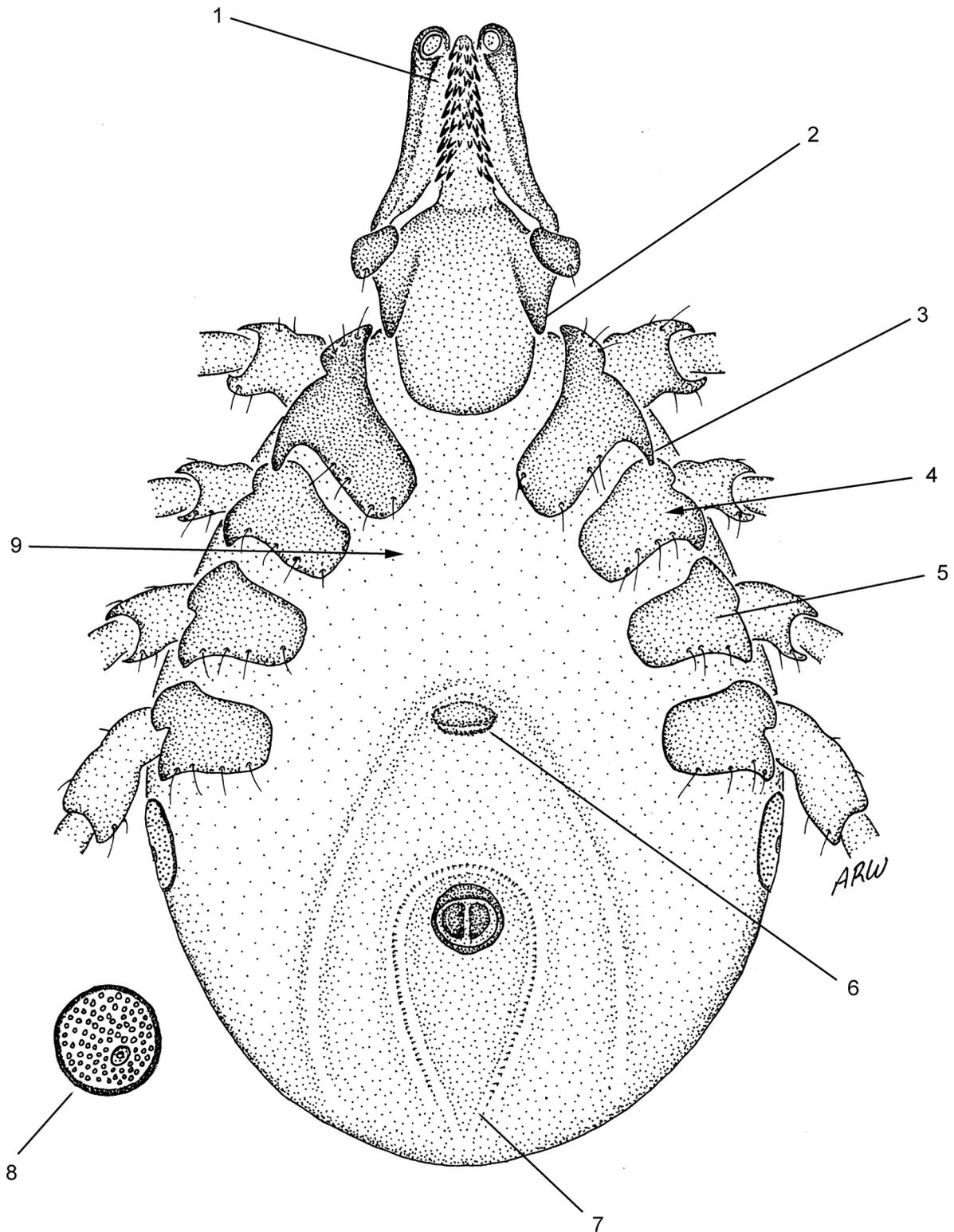
The following refers only to adult females (Figs 45 & 46). *Ixodes cornuatus* can be distinguished easily from *I. hirsti* and *I. tasmani* by the presence of cornua at the posterior margin of the basis capituli and the presence of corrugations at the posterior margins of the scutum in *I. cornuatus* but not on *I. hirsti* nor *I. tasmani*. Also, *I. cornuatus* has long cervical grooves on the scutum whereas these are short on *I. hirsti*. And the punctations on the scutum of *I. cornuatus* are sparse and small compared to the dense and large punctations of *I. tasmani*.



- 1 Palp article 1 is bulbous and distant from cheliceral sheath.
- 2 Palp shape is long and slender.
- 3 Basis dorsal posterior margin is undulating.
- 4 Cornua are present.
- 5 Cervical grooves are long.
- 6 Punctation size on scutum is small.

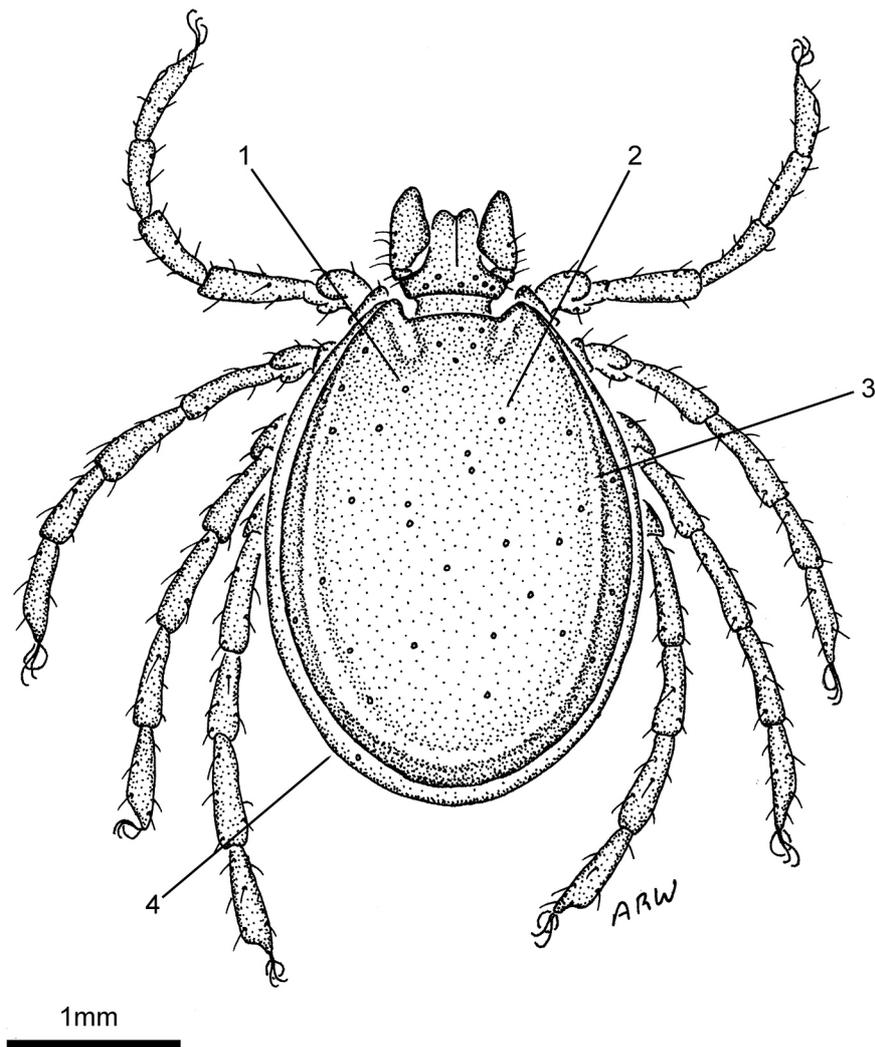
- 7 Punctation distribution on scutum is sparse.
- 8 Scutal corrugations at posterior are present.
- 9 Posterior grooves are absent.
- 10 Tarsi distal profile is steeply stepped.
- 11 Leg colour pattern is absent.
- 12 Scutal proportion is longer than wide.
- 13 Lateral carinae are present.

FIGURE 45. *Ixodes cornuatus* (southern paralysis tick), female, dorsal.



- | | |
|---------------------------------------|---|
| 1 Hypostome shape narrows anteriorly. | 6 Genital aperture position is level with coxae 4. |
| 2 Auriculae are present. | 7 Anal groove shape is joined at posterior to form a point. |
| 3 Coxae 1 external spur is present. | 8 Spiracle plate shape is circular. |
| 4 Coxa type is without syncoxae. | 9 Sternal plate is absent. |
| 5 Coxae surface texture is smooth. | |

FIGURE 46. *Ixodes cornuatus* (southern paralysis tick), female, ventral.



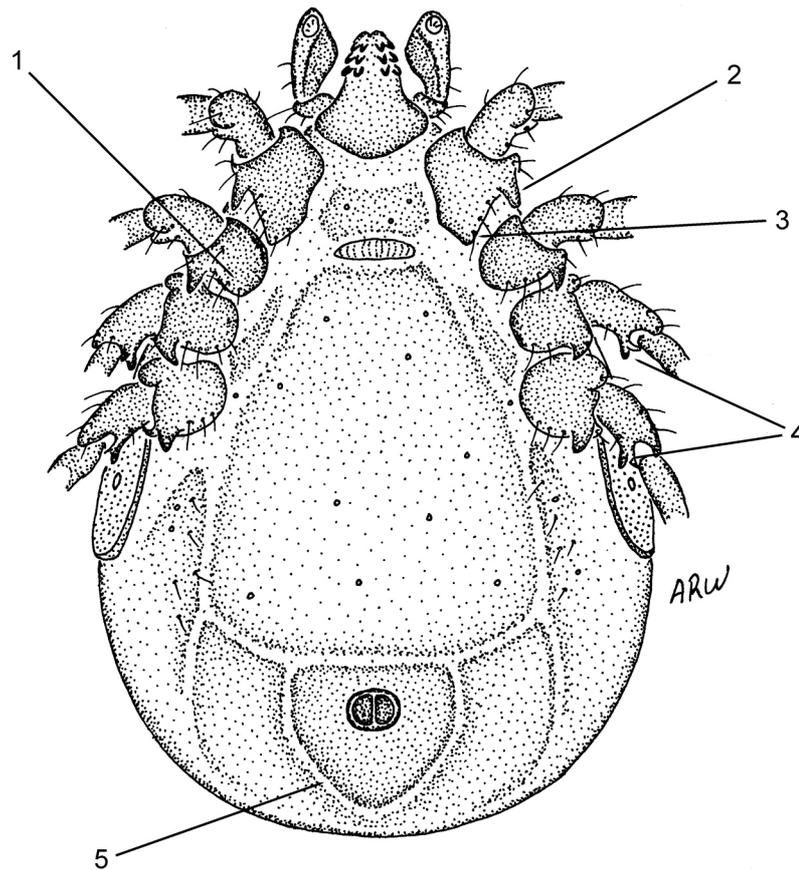
- 1 Punctuation size on conscutum is small.
- 2 Punctuation distribution on conscutum is sparse.
- 3 Lateral groove on conscutum is present.
- 4 Body profile is a broad oval.

FIGURE 47. *Ixodes cornuatus* (southern paralysis tick), male, dorsal.

To distinguish *I. cornuatus* from *I. holocyclus* in areas where these species might overlap (Figs 49 & 58) the following characters should be compared. *Ixodes cornuatus* has distinct cornua whereas *I. holocyclus* does not have cornua, or the cornua are indistinct. *Ixodes cornuatus* has a scutum longer than wide; the scutum tends to have a concave profile to both sides of the posterior scutum. *Ixodes holocyclus* has a scutum wider than long, and if there is any concavity at the posterior sides of the scutum it is slight. The legs of females of *I. cornuatus* are all the same brown colour, compared to *I. holocyclus* which has paler second and third pairs of legs.

Hosts.

There is uncertainty about the records of *I. cornuatus* from mainland Australia since it has been difficult to distinguish unequivocally *I. cornuatus* and *I. holocyclus*, until now, in our opinion. Nonetheless, we list all of the putative hosts of *I. cornuatus* here. [Only *I. cornuatus* is known from Tasmania so we are confident about all of the Tasmania records of *I. cornuatus*.]



- 1 Coxae type is without syncoxae.
- 2 Coxae external spurs are present.
- 3 Coxae internal spurs are absent.
- 4 Trochanter spurs on legs 3 and 4 are distinct.
- 5 Anal groove shape is joined at posterior.

FIGURE 48. *Ixodes cornuatus* (southern paralysis tick), male, ventral.

Host-records from Roberts (1960, 1964b, 1970): *Isodon obesulus* (southern brown bandicoot), *Thylogale billardierii* (Tasmania pademelon), *Trichosurus vulpecula fuliginosus* (common brushtail possum), *Bettongia gaimardi cuniculus* (Tasmania bettong), *Perameles gunnii* (eastern barred bandicoot), *Rattus rattus* (black rat), *R. lutreolus* (swamp rat), *Mus musculus* (house mouse), *Strepera fuliginosa* (black currawong), *Cracticus torquatus* (grey butcherbird), *Colluricincla harmonica* (grey shrike-thrush) and *Sericornis humilis* (Tasmanian scrubwren). *Phascolarctos cinereus* (koala), *Vombatus ursinus* (common wombat), *Macropus robustus* (common wallaroo), *Homo sapiens* (human), *Canis familiaris* (domestic dog, introduced), *Felis catus* (domestic cat, introduced), *Bos taurus* (domestic cattle, introduced), and *Ovis aries* (domestic sheep, introduced).

Host-records since Roberts (1970) (from two different papers): (i) *Vombatus ursinus* (common wombat, Moorhouse & Heath, 1975); and (ii) *Rattus fuscipes* (bush rat), *Vombatus ursinus* (common wombat), *Felis catus* (domestic cat, introduced) and *Canis familiaris* (domestic dog, introduced) (Jackson *et al.* 2007).

Life-cycle and seasonality.

Nothing is known of the life-cycle or seasonality of *I. cornuatus*, but its life-cycle might be similar to that of its close relative *I. holocyclus* (refer to the species accounts of *I. holocyclus*).



FIGURE 49. Approximate geographic distribution of *Ixodes cornuatus* (southern paralysis tick) (redrawn from Kelers *et al.* 2012).

Disease.

Ixodes cornuatus may cause paralysis in dogs (Roberts 1960, Mason *et al.* 1974, Beveridge *et al.* 2004), cats (Mason *et al.* 1974) and sheep (Sloan 1968). Tick paralysis caused by *I. cornuatus* has not been compared, in detail, to tick paralysis caused by *I. holocyclus* but may be similar. Thus, the reader is referred to our species account of *I. holocyclus*.

Habitat and geographic distribution.

Distinguishing female *I. cornuatus* from *I. holocyclus* by their morphology has been exceptionally difficult in the past. Indeed, the only unequivocal method has been to compare the nucleotide sequences of *cytochrome oxidase* (Song *et al.* 2011). We hope that characters 4, 5, 7, 9, 10, 11 & 12 of Figs 45 and 54 and will help future workers. We do not feel confident about the mainland localities of *I. cornuatus* except for the specimens identified by Roberts (Roberts 1960, 1970 & Sloan 1968) and Beveridge *et al.* (2004), and indeed any of the localities of *I. holocyclus* from Victoria and southeastern New South Wales except for those identified by Roberts (1960, 1970) and Beveridge (1991). We are not confident about the localities of Jackson *et al.* (2007) since we have not been able to reconcile the genetic and morphological criteria of Jackson *et al.* (2000, 2002) with the genetic criteria of Song *et al.* (2011) and our new morphological criteria (the present work). Thus, for us, the only localities of *I.*

cornuatus in Victoria of which we are confident are: (i) Orbost (Lake Tyers) (Sloan, 1968); (ii) Warragul District, Noojee Neerim North, and Lakes Entrance (Roberts 1960); (iii) Dandenong, Mt Buffalo, and Bairnsdale near Lakes Entrance (Roberts 1970); (iv) Daylesford and Leongatha (Beveridge *et al.* 2004); and (v) Mallacoota, Orbost, Bullengarook, Silvan and Donvale (Song *et al.* 2011). Also, for us the only locality of *I. cornuatus* in New South Wales of which we are confident is Brownlee (Roberts 1960).

Roberts identified both *I. cornuatus* and *I. holocyclus* in a batch of ticks from Lake Tyers, Orbost "in approximately equal numbers"; these words are intriguing. It would be very interesting if indeed two very similar species were present at one site in approximately equal numbers. It would also be interesting to know if there are any *I. cornuatus* - *I. holocyclus* hybrids at Lake Tyers, Orbost. Song *et al.* (2011) also found both *I. cornuatus* and *I. holocyclus* at Orbost. Arundel, cited by Bagnall & Doube (1975), stated that *I. cornuatus* is particularly common in the Central Highlands of Victoria but this needs to be confirmed.

Virtually nothing is known about the preferred habitat of *I. cornuatus*. Indeed, very little is known about the biology of this tick despite the fact that it causes life-threatening disease in dogs and other domestic animals and it is the most common tick that affects livestock in Tasmania (Arundel & Sutherland 1988).

Genes and genomes.

Parts of the following genes are available to the public in GenBank: (i) Internal Transcribed Spacer (ITS2) rRNA (568 bp, Song *et al.* 2011; Shaw *et al.* 2002); (ii) *cytochrome c oxidase subunit I (cox1)* (644 bp, Song *et al.* 2011); and (iii) small rRNA subunit, control region #1, tRNA-Ile, tRNA-Gln, tRNA-Met (Shao *et al.* 2005a).

Other information.

None.

Ixodes hirsti Hassall, 1931 (Hirst's marsupial tick)

General

Ixodes hirsti is known as Hirst's marsupial tick. *Ixodes hirsti* was named after Stanley Hirst, Department of Zoology, University of Adelaide, South Australia. *Ixodes hirsti* is one of 28 species in the Australasian lineage of the genus *Ixodes* (Barker & Murrell 2004, 2008) and one of seven species in the subgenus *Sternalixodes*, all of which occur only in Australia, except for *I. confusus* and *I. cordifer* which also occur in Papua New Guinea.

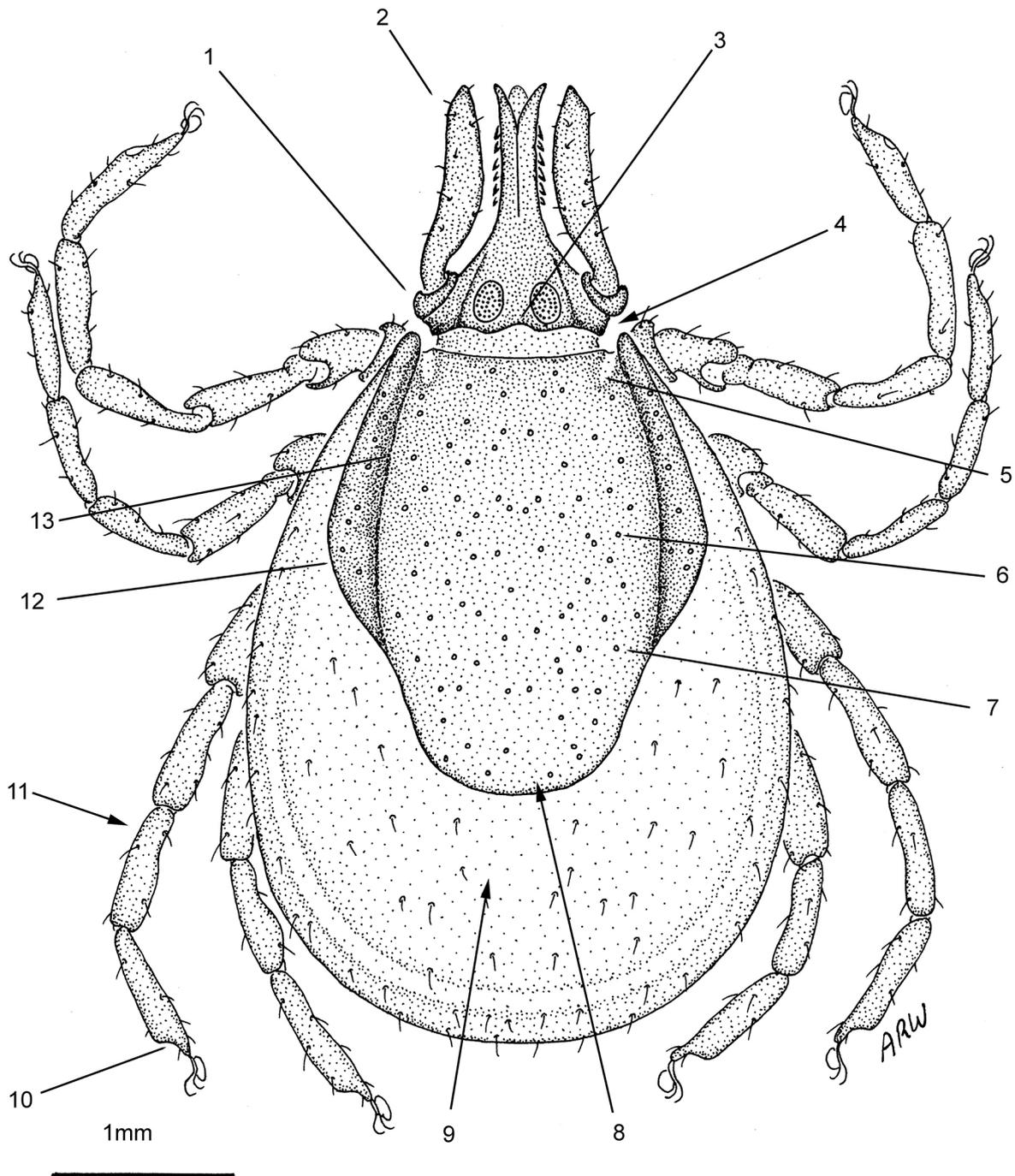
Differential diagnosis

The following refers only to adult females (Figs 50 & 51). *Ixodes hirsti* is distinguished from *I. cornuatus* by the absence of cornua on the posterior margin of the basis capituli, and by the absence of corrugations at the posterior of the scutum. *Ixodes hirsti* has short cervical grooves whereas the cervical grooves in *I. cornuatus* and *I. tasmani* are long. Punctations on the scutum of *I. hirsti* are sparse and small compared to the dense and large punctations of *I. tasmani*. *Ixodes hirsti* has both a scutum longer than wide, and sides of the posterior scutum that are concave, in comparison to those of *I. holocyclus* and *I. tasmani*. The larvae and a putative nymph of *I. hirsti* were described and illustrated with drawings and electron micrographs by Laan *et al.* (2011b).

Hosts

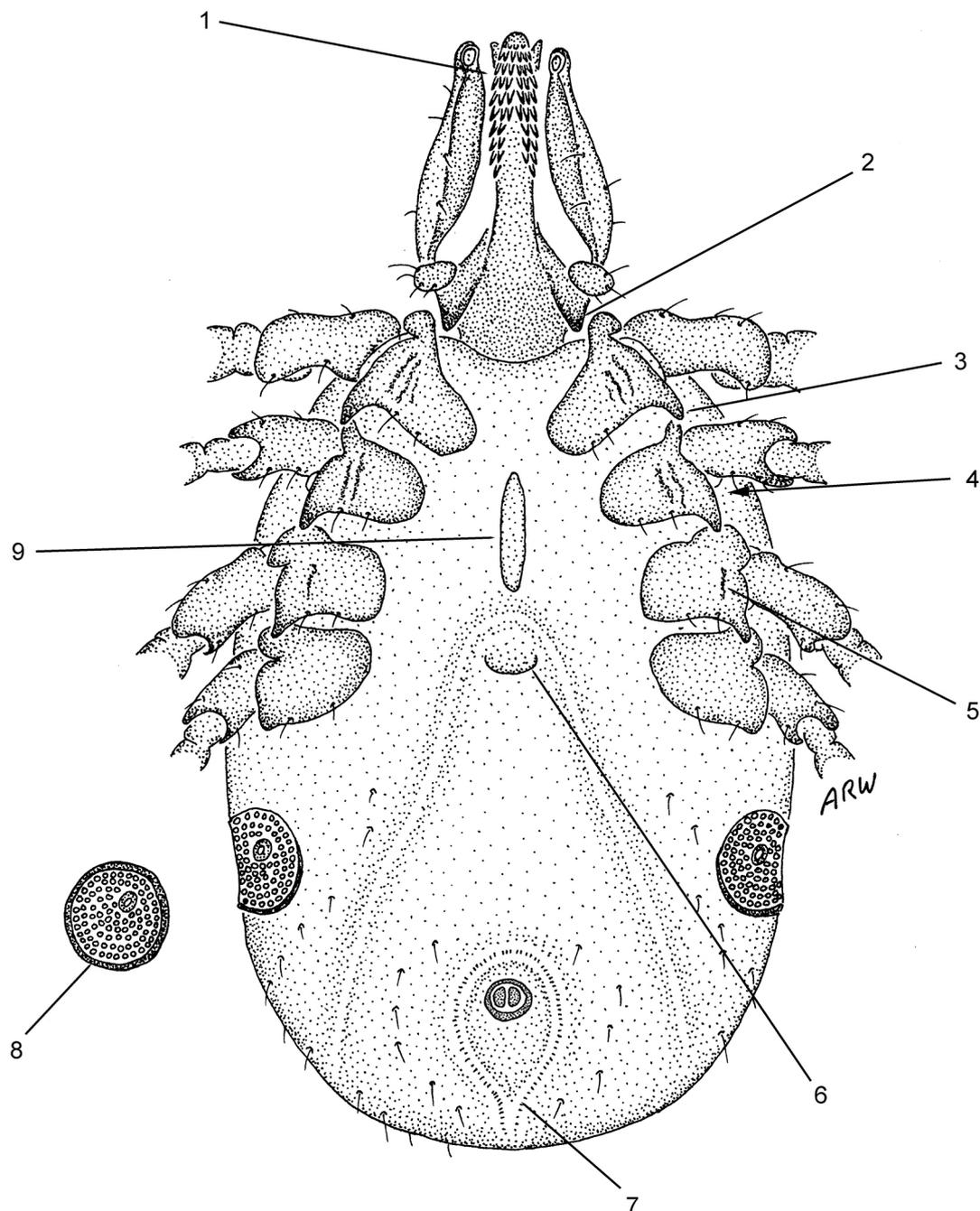
Ixodes hirsti is far less catholic in its feeding habits than the other *Ixodes* species in this book: *I. cornuatus*, *I. holocyclus* and *I. tasmani*. *Ixodes hirsti* has been recorded from only about 10 species of Australian marsupials (possums; kangaroos and their kin), domestic dogs and cats and some birds. Nonetheless, *I. hirsti* is a common tick of mammals and, in the nymphal and larval stages, of birds in southern Australia (Laan *et al.* 2011b & references therein). Birds that descend to the ground to forage and drink are thought to be the most common hosts of larval *I. hirsti* (Oorebeek & Kleindorfer, 2008a).

Host-records from Roberts (1970): *Trichosurus caninus* (short-eared brushtail possum), *T. vulpecula fuliginosus* (Tasmanian brush-tailed possum), *Phascolarctos cinereus* (koala), *Potorous tridactylus* (long-nosed potoroo), *Wallabia bicolor* (swamp wallaby), *Macropus giganteus* (eastern grey kangaroo), *Rattus fuscipes* (bush rat), *R. rattus* (black rat, introduced), and *Felis catus* (domestic cat, introduced).



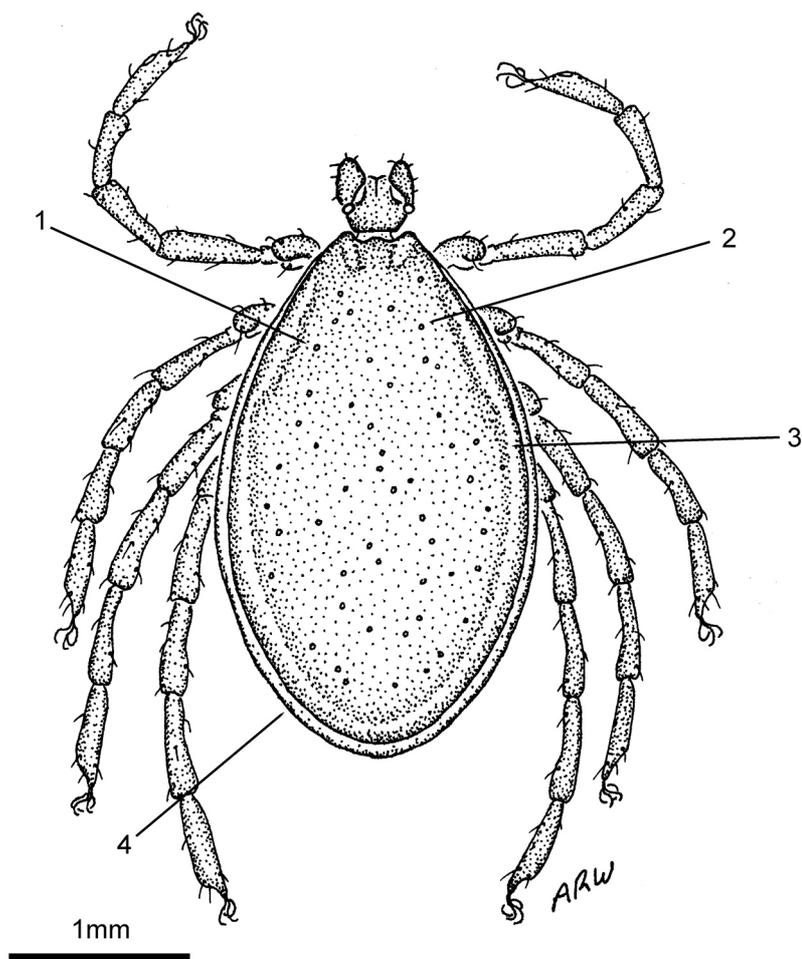
- | | |
|--|---|
| <p>1 Palp article 1 is bulbous and distant from the cheliceral sheath.</p> <p>2 Palps shape is long and slender.</p> <p>3 Basis dorsal posterior margin is undulating.</p> <p>4 Cornua are absent.</p> <p>5 Cervical grooves are short.</p> <p>6 Punctuation size on scutum is small.</p> <p>7 Punctuation distribution on scutum is sparse.</p> | <p>8 Scutal corrugations at posterior are absent.</p> <p>9 Posterior grooves are absent.</p> <p>10 Tarsi distal profile is steeply stepped (on legs 3 and 4).</p> <p>11 Leg colour pattern is absent.</p> <p>12 Scutum proportion is longer than wide.</p> <p>13 Lateral carinae are present.</p> |
|--|---|

FIGURE 50. *Ixodes hirsti* (Hirst's marsupial tick), female, dorsal.



- 1 Hypostome shape narrows anteriorly.
- 2 Auriculae are present.
- 3 Coxae 1 external spur is present.
- 4 Coxae type is without syncoxae.
- 5 Coxae surface texture is ridged (coxae 1 to 3).
- 6 Genital aperture position is level with coxae 4.
- 7 Anal groove shape is joined at posterior to form a point.
- 8 Spiracle plate shape is circular.
- 9 Sternal plate is present.

FIGURE 51. *Ixodes hirsti* (Hirst's marsupial tick), female, ventral.



- 1 Punctuation size on conscutum is small.
- 2 Punctuation distribution on conscutum is sparse.
- 3 Lateral groove on conscutum is present.
- 4 Body profile is a narrow oval.

FIGURE 52. *Ixodes hirsti* (Hirst's marsupial tick), male, dorsal.

Host-records since Roberts (1970) (from two different papers): (i) records of adult *Ixodes hirsti* from publications and museum specimens, compiled by Laan *et al.* (2011b, Table 1); (ii) *Macropus fuliginosus* (western grey kangaroo) (Oorebeek & Rismiller 2007); and (iii) *M. giganteus* (eastern grey kangaroo), *M. rufogriseus* (red-necked wallaby), *Petrogale penicillata* (brush-tailed rock wallaby), *Thylogale thetis* (red-necked pademelon), *Wallabia bicolor* (swamp wallaby), *Bettongia penicillata* (brush-tailed bettong), *Pseudocheirus peregrinus* (common ringtail possum), *Phascolarctos cinereus* (koala), *Canis familiaris* (domestic dog, introduced), *Felis catus* (domestic cat, introduced), *Phylidonyris novaehollandiae* (New Holland honeyeater), *P. pyrrhoptera* (crescent honeyeater), *P. melanops* (tawny-crowned honeyeater), *Melithreptus brevirostris* (brown-headed honeyeater), *Acanthiza lineata* (striated thornbill), *A. pusilla* (brown thornbill), *Sericornis frontalis* (white-bowed scrubwren), *Pardalotus punctatus* (spotted pardalote), and *Petroica multicolour* (scarlet robin) (Chapman *et al.* 2009).

Life-cycle and seasonality

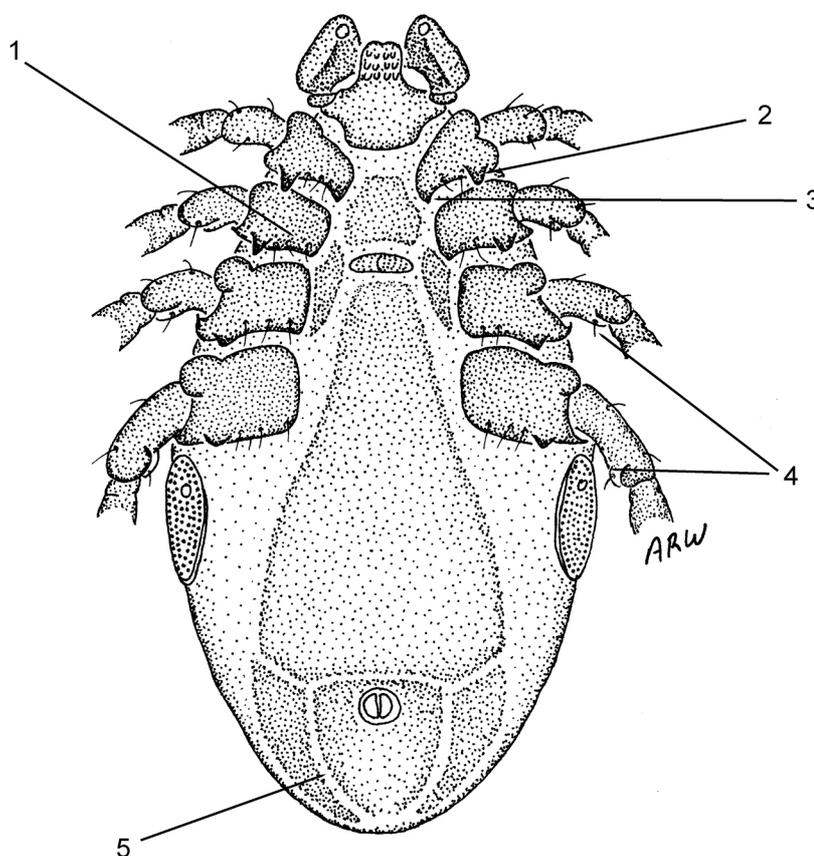
Ixodes hirsti is a three-host tick as are all of the 243 species of *Ixodes* that have been studied so far. Adult male and female *I. hirsti* infect mammals whereas the nymphs, and particularly the larvae infect birds (Laan *et al.* 2011b and references therein). In one study of *I. hirsti* on *Phascolarctos cinereus* (koala) over three years, 186 ticks were collected (Laan *et al.* 2011b). One hundred and sixty of these ticks (86%) were adult females, 22 (12%) were adult males and four were nymphs; no larvae were collected. Most of these ticks (74%) were found on the head, neck and

forearms of their hosts. None of the 22 adult male ticks were attached to their hosts but these males were usually found next to females that were attached to their hosts. Larvae were found on vegetation primarily in September and October (one year sample) whereas adults were found on hosts for just six months of each year (August to January, three year sample).

Disease

Roberts (1961) reported paralysis in two domestic cats in South Australia but there have not been any further reports of paralysis caused by *I. hirsti* in the scientific literature. Furthermore, Arundel (1984) pointed out that pathological tests were not done to exclude other diseases in the cats studied by Roberts (1961). Our null hypothesis is that *I. hirsti* does not cause paralysis in cats nor other animals. Future cases of paralysis in domesticated animals or wildlife caused by *I. hirsti*, or indeed any Australian tick, would be most worthy of a report in the scientific literature. Kemp (1979) proposed that all seven species in the subgenus *Sternalixodes* may cause paralysis in their hosts: *I. holocyclus*, *I. cornuatus*, *I. confusus*, *I. cordifer*, *I. hirsti*, *I. myrmecobii* and *I. trichosuri*.

Larval *I. hirsti* are common on passerine birds, at least in South Australia (Oorebeek & Kleindorfer 2008a) where this tick has been studied most. It is not known to what extent larval and nymphal *I. hirsti* harm their bird-hosts.



- 1 Coxae type is without syncoxae.
- 2 Coxae external spurs are present.
- 3 Coxae internal spurs are present (on coxae 1).
- 4 Trochanter spurs on legs 3 and 4 are indistinct.
- 5 Anal groove shape is joined at posterior.

FIGURE 53. *Ixodes hirsti* (Hirst's marsupial tick), male, ventral.

Habitat and geographic distribution

Ixodes hirsti is a tick of dense understory vegetation of the sub-coastal areas of southern Australia (Oorebeek & Kleindorfer 2008b, Laan *et al.* 2011b).

Genes and genomes

Chapman *et al.* (2009) studied the genetic structure of larvae and nymphs on Kangaroo Island, South Australia and Fleurieu Peninsula, mainland South Australia, with mitochondrial DNA sequences (*cytochrome oxidase I*, 12S rRNA, control region #1, tRNA-Ile, tRNA-Gln and tRNA-Met 491bp). The ticks studied could not be identified to species with confidence, however, at the time of the study. Subsequently, however, Laan *et al.* (2011b) described the larvae of *I. hirsti* Hassall, 1931 so the ticks studied by Chapman *et al.* (2009) could now be identified to species, retrospectively, with confidence. Shao *et al.* (2005a) studied the following mitochondrial genes: tRNA-Leu, control region #2, large subunit (16S) rRNA (656bp), and small subunit (12S) rRNA, control region #1, tRNA-Ile, tRNA-Gln, tRNA-Met (792 bp).

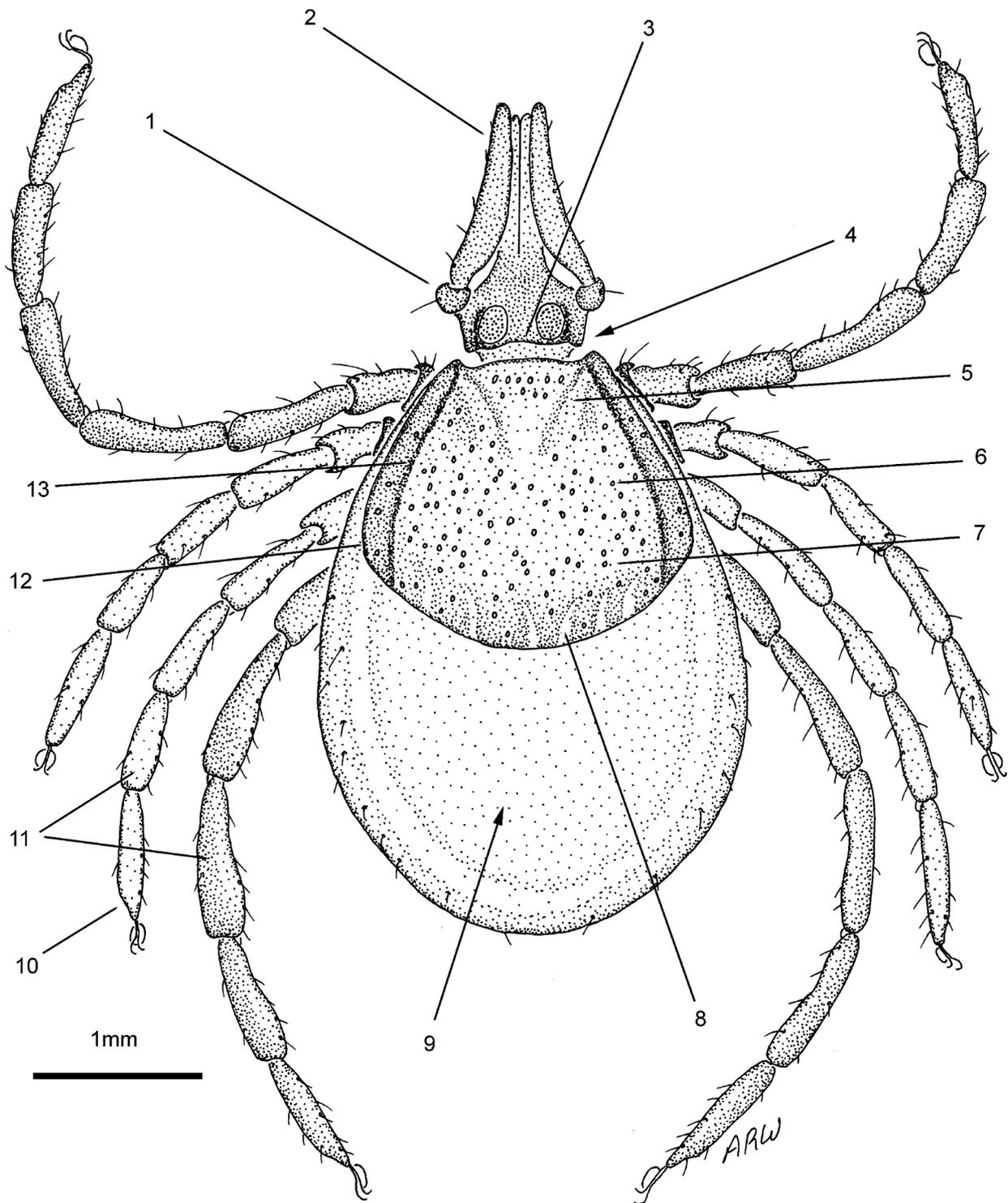
Other information

Kleindorfer *et al.* (2006), Oorebeek & Kleindorfer (2008a, 2008b) and Oorebeek *et al.* (2009) are elegant papers on the ecology of the immature stages, particularly the larvae of a tick that the authors concluded was likely to be *Ixodes hirsti* Hassall, 1931. The identity of this tick could not be determined for certain at the time of the studies but subsequently Laan *et al.* (2011b) described the larvae of *I. hirsti*. Thus, the larvae studied by Kleindorfer *et al.* (2006), Oorebeek & Kleindorfer (2008a, 2008b) and Oorebeek *et al.* (2009) could be identified to species, retrospectively, with certainty. Kleindorfer *et al.* (2006) found larvae and nymphs of *I. hirsti* (species identity to be confirmed) on 22 of 126 (17.5%) *Phylidonyris novaehollandiae* (New Holland honeyeaters). The prevalence of ticks on this host was statistically significantly higher in juvenile compared to adult birds in two coastal areas compared to two mountain areas, and in birds with low body condition scores compared to birds with high body condition scores. Oorebeek & Kleindorfer (2008a) found larval and nymphal *I. hirsti* (species identity to be confirmed) on passerine birds on Kangaroo Island, South Australia, for 8 months of each of two years: April to November. The intensity of infection was highest in June, July and August, the winter months. Tick prevalence and tick intensity were greatest from June to September, the breeding season of the main hosts of *I. hirsti* in this study, honeyeaters, scrubwrens and thornbills. Oorebeek & Kleindorfer (2008b) found that the number of shrubs and the percent ground-cover by shrubs were statistically significant predictors of the presence of *I. hirsti* (species identity to be confirmed) at six sites in SA. Oorebeek *et al.* (2009) found that in the laboratory larval *I. hirsti* (species identity to be confirmed) were attracted to shade away from direct sunlight. This was interpreted as an adaptation to conserve water. Larvae were also attracted to heat but not host odour nor carbon dioxide. This was interpreted as evidence for an ambush strategy by the ticks for their hosts. The average questing height of *I. hirsti* in the laboratory was just 12.4 cm.

***Ixodes holocyclus* Neumann, 1899 (paralysis tick)**

General

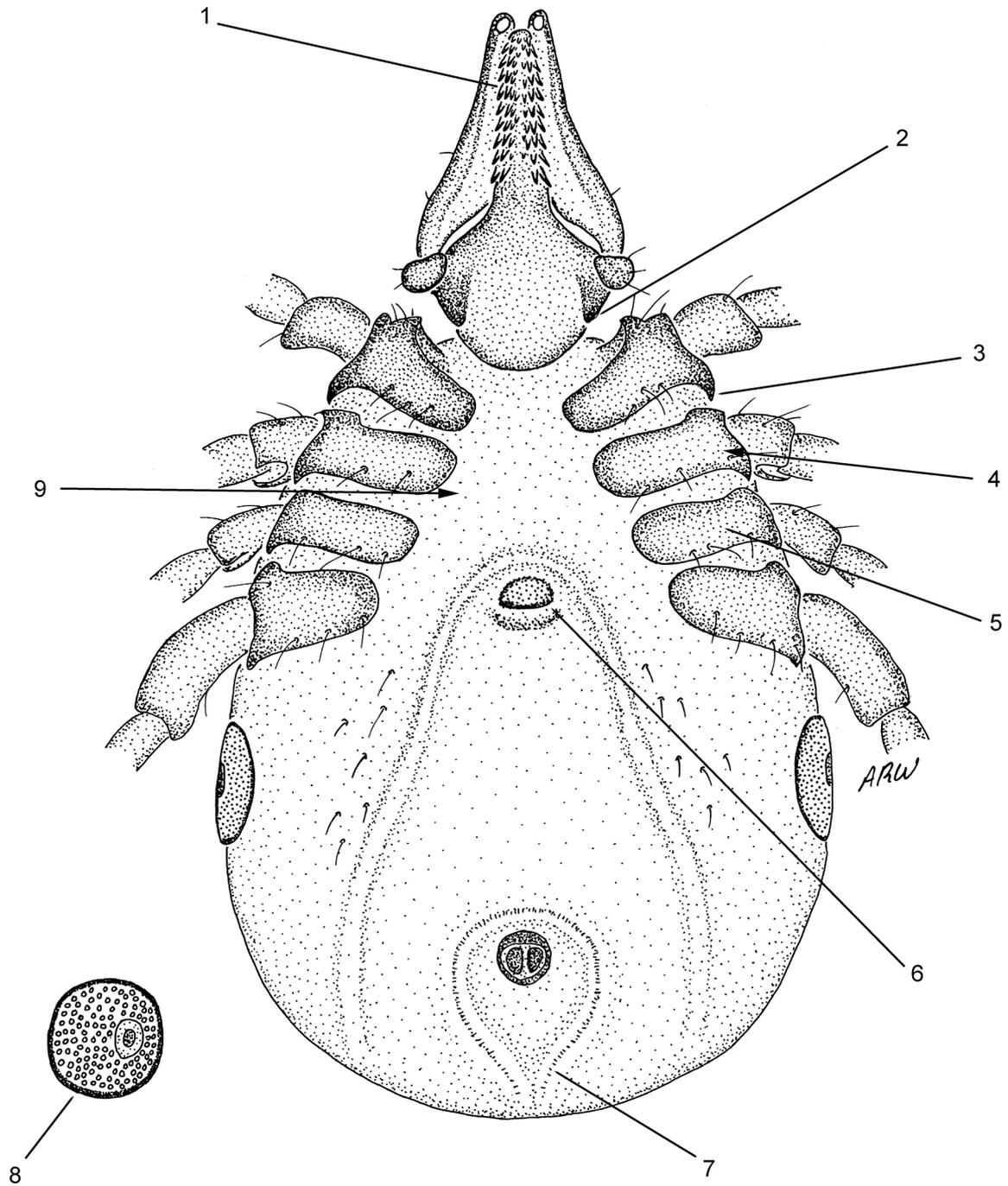
Ixodes holocyclus is known as the Australian paralysis tick since most cases of tick paralysis in domestic animals, wildlife and humans in Australia are caused by this species of tick. *Ixodes holocyclus* is also known as the scrub tick in Queensland, particularly in north and far north Queensland. The name scrub tick echoes the predilection of *I. holocyclus* for forested areas, particularly wet forested areas, which are often referred to as "scrub" in north and far north Queensland. *Ixodes holocyclus* is one of 28 species in the Australasian lineage of the genus *Ixodes* (Barker & Murrell 2004, 2008) and one of seven species in the subgenus *Sternalixodes*, all of which occur only in Australia, except for *I. confusus* and *I. cordifer* which also occur in Papua New Guinea.



- 1 Palp article 1 is bulbous and distant from cheliceral sheath.
- 2 Palp shape is long and slender.
- 3 Basis dorsal posterior margin is undulating.
- 4 Cornua are absent.
- 5 Cervical grooves are short.
- 6 Punctation size on scutum is small.
- 7 Punctation distribution on scutum is dense.

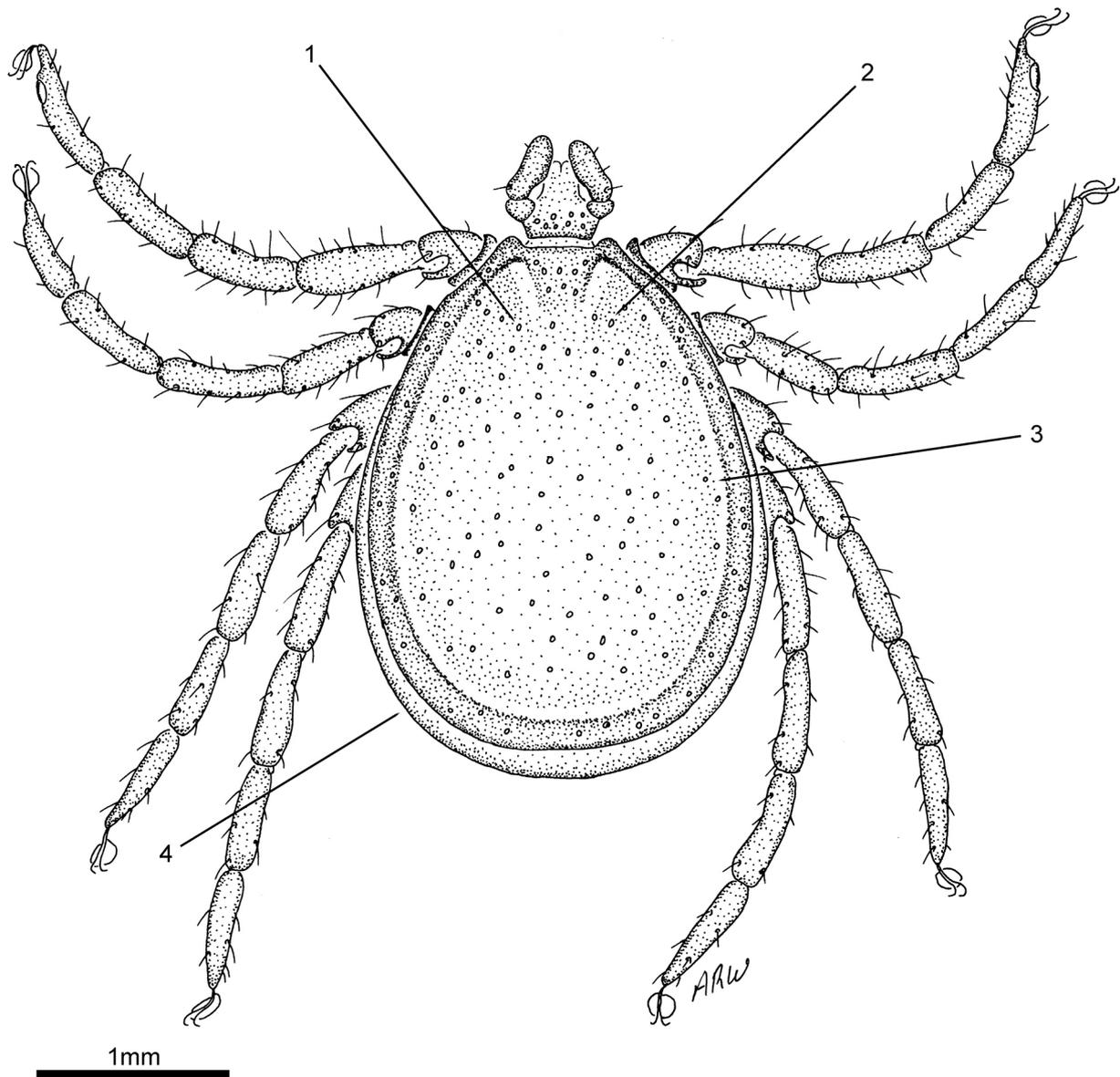
- 8 Scutal corrugations at posterior are present.
- 9 Posterior grooves are absent.
- 10 Tarsi distal profile is gradually stepped.
- 11 Leg colour pattern is legs 1 and 4 darker than legs 2 and 3.
- 12 Scutal proportion is wider than long.
- 13 Lateral carinae are present.

FIGURE 54. *Ixodes holocyclus* (paralysis tick), female, dorsal.



- | | |
|---------------------------------------|---|
| 1 Hypostome shape narrows anteriorly. | 6 Genital aperture position is level with coxae 4. |
| 2 Auriculae are present. | 7 Anal groove shape is joined at posterior to form a point. |
| 3 Coxae 1 external spur is present. | 8 Spiracle plate shape is circular. |
| 4 Coxae type is without syncoxae. | 9 Sternal plate is absent. |
| 5 Coxae surface texture is smooth. | |

FIGURE 55. *Ixodes holocyclus* (paralysis tick), female, ventral.



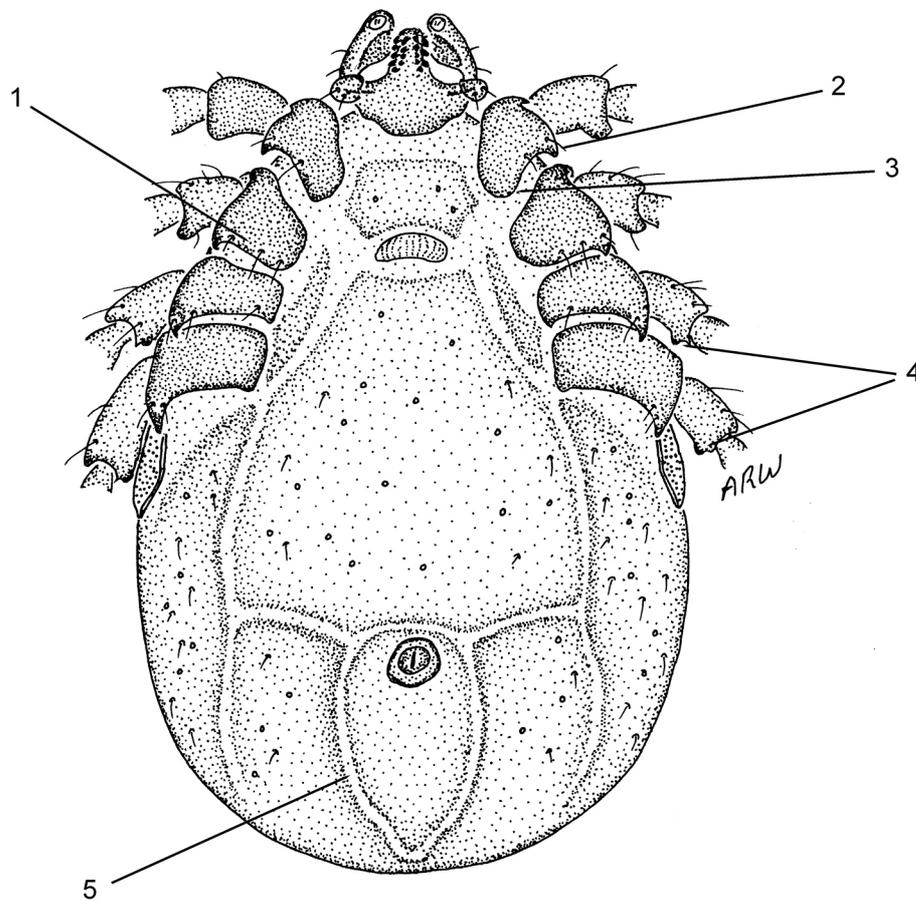
- 1 Punctuation size on conscutum is small.
- 2 Punctuation distribution on conscutum is dense.
- 3 Lateral groove on conscutum is present.
- 4 Body profile is a broad oval.

FIGURE 56. *Ixodes holocyclus* (paralysis tick), male, dorsal.

Differential diagnosis

Ixodes holocyclus is very closely related, and thus morphologically similar, to *I. cornuatus* and *I. myrmecobi*. *Ixodes myrmecobi* is only known from southwestern Western Australia where it infests the marsupial numbat, *Myrmecobius fasciatus*. Veterinarians, medical workers and scientists alike have been baffled by the morphology of *I. holocyclus* and *I. cornuatus* for most of the last century. But detailed studies since 2000 have resolved the stated and unstated questions about *I. holocyclus* and *I. cornuatus* (Jackson *et al.* 2000, 2002; Shaw *et al.* 2002; Song *et al.* 2011). Thus we may conclude that: (i) *I. holocyclus* and *I. cornuatus* are genetically distinct and thus warrant the species-status given to them by Roberts (1960, 1970); and (ii) female *I. holocyclus* and *I. cornuatus* may be distinguished morphologically (compare Figs 54 & 55 to Figs 45 & 46). Fortunately, all stages (larvae,

nymphs, males and females) of *I. holocyclus* may be distinguished unambiguously from those of *I. cornuatus* by comparison of mitochondrial (*cytochrome oxidase I*) and nuclear (*ITS2*) nucleotide sequences (Song *et al.* 2011, Kelers *et al.* 2012). Also, Jackson *et al.* (2002) proposed that larvae of *I. holocyclus* and *I. cornuatus* can be distinguished by the numbers and lengths of setae.



- 1 Coxae type is without syncoxae.
- 2 Coxae external spurs are present.
- 3 Coxae internal spurs are absent.
- 4 Trochanter spurs of legs 3 and 4 are indistinct.
- 5 Anal groove shape is joined at posterior.

FIGURE 57. *Ixodes holocyclus* (paralysis tick), male, ventral.

The following applies only to morphological features of adult females (Figs 16, 54 & 55). *Ixodes holocyclus* has the unusual feature for ticks of the first and fourth pairs of legs being darker in general colour (brown) compared to the second and third pairs has of legs (beige). This character-state is not seen in the other *Ixodes* species in this book. *Ixodes holocyclus* can be distinguished from *I. tasmani* by the sparse and small punctations on its scutum, its short cervical grooves and corrugations at the posterior scutum, compared to punctations that are dense and large, with long cervical grooves, and no corrugations at the posterior scutum in *I. tasmani*. *Ixodes holocyclus* can be distinguished from *I. hirsti* by the its scutum being wider than long, and corrugations at the posterior scutum; compared to the scutum being longer than wide, and no corrugations at the posterior scutum in *I. hirsti*. To differentiate *I. holocyclus* from *I. cornuatus* in areas where both species might occur (Figs 49 & 58), the cornua and proportions of the scutum should be compared. *Ixodes holocyclus* has no, or possibly indistinct, cornua at the posterior margin of the basis capituli whereas *I. cornuatus* has distinct cornua. The scutum of *I. holocyclus* is wider

than long; the scutum has no, or only a slight, concave profile of the sides of the posterior part of the scutum, compared to *I. cornuatus* which has a longer than wide scutum, and a concave profile of the sides of the posterior part of the scutum.



FIGURE 58. Approximate geographic distribution of *Ixodes holocyclus* (paralysis tick) (redrawn from Seddon 1968).

Hosts

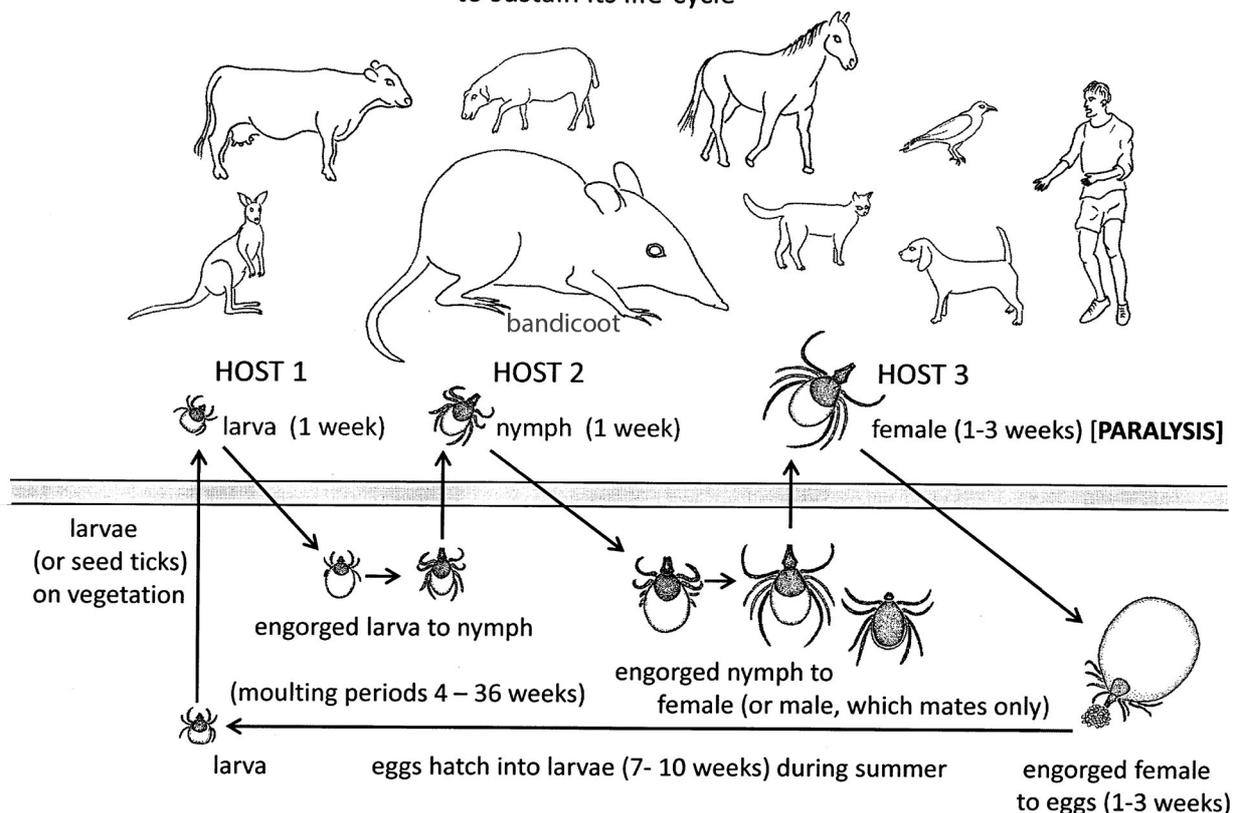
Ixodes holocyclus is catholic in its feeding habits. Indeed, *I. holocyclus* has been recorded from 34 species of mammals and seven species of birds (below). Where *I. holocyclus* is abundant, it will be found on most of the species of mammals present, but the bandicoots *Isodon macrourus* and *Perameles nasuta* have been considered the principal hosts in southeastern Queensland since at least 1975 (Doube 1975e). These bandicoots may carry many ticks. Doubtless, reasonable numbers of *I. macrourus* and *P. nasuta* are required for populations of *I. holocyclus* to persist from one tick season to another in southeastern Queensland but it is unknown if this is the case in other parts of the geographic range of *I. holocyclus*.

Host-records from Roberts (1970): *Tachyglossus aculeatus* (short-beaked echidna), *Antechinus flavipes* (yellow-footed antechinus), *Phascogale tapoatafa* (brush-tailed phascogale), *Sminthopsis murina* (common dunnart), *Perameles nasuta* (long-nosed bandicoot), *Isodon macrourus* (northern brown bandicoot), *I. obesulus* (southern brown bandicoot), *Trichosurus caninus* (short-eared brushtail possum), *T. vulpecula* (common brushtail possum), *Phascolarctos cinereus* (koala), *Aepyprymnus rufescens* (rufous bettong), *Thylogale stigmatica* (red-legged pademelon), *Wallabia bicolor* (swamp wallaby), *Macropus dorsalis* (black-striped wallaby), *Macropus*

rufogriseus (red-necked wallaby), *Dendrolagus lumholtzi* (Lumholtz's tree kangaroo), *Homo sapiens* (man), *Oryctolagus cuniculus* (wild rabbit, introduced), *Melomys cervinipes* (fawn-footed melomys), *Uromys caudimaculatus* (giant white-tailed rat), *Mus domesticus* (house mouse, introduced), *Rattus sordidus* (canefield rat), *R. tunneyi* (pale field-rat), *R. fuscipes* (bush rat), *R. norvegicus* (brown rat, introduced), *R. rattus* (black rat, introduced), *Hydromys chrysogaster* (water rat), *Canis familiaris* (domestic dog, introduced), *C. lupus* (dingo), *Felis catus* (domestic cat, introduced), *Equus caballus* (domestic and feral horses, introduced), *Sus scrofa* (feral pig, introduced), *Capra hircus* (domestic goat, introduced), and the birds, *Gallus gallus* (domestic fowl, introduced), *Meleagris gallopavo* (domestic turkey, introduced), *Platycercus elegans* (crimson rosella), *Pitta* sp. (pitta), *Corvus coronoides* (Australian raven), *Cracticus nigrogularis* (pied butcher-bird) and *Cracticus tibicen* (Australian magpie).

Host-records since Roberts (1970) (from seven different sources): (i) *Canis lupus* (dingo), *Trichosurus vulpecula* (common brushtail possum), *Cracticus nigrogularis* (pied butcher bird) and *Cracticus tibicen* (Australian magpie) (Marks & Cribb 1966); (ii) *Trichosurus canis* (short-eared brushtail possum) (Presidente *et al.* 1982); (iii) *Dactylopsila trivirgata* (striped possum) (Jackson 1998); (iv) *Petrogale penicillata* (brush-tailed rock-wallaby) (Barnes *et al.* 2010); (v) *Pteropus conspicillatus* (spectacled flying fox) (Buettner *et al.* 2013); (vi) *Phascolarctos cinereus* (koala) (Stone & Carrick, 1990); and (vii) *Petrogale godmani* (Godman's rock-wallaby), *I. macrourus* (northern brown bandicoot), *Bettongia tropica* (northern bettong), *Dendrolagus lumholtzi* (Lumholtz's tree kangaroo), *Canis familiaris* (domestic dog, introduced), *Bos taurus* (domestic cattle, introduced) and *Pteropus conspicillatus* (spectacled flying fox) (Barker & Campelo unpublished data).

FEEDING (on host) *Ixodes holocyclus* feeds on any of these hosts but needs bandicoots to sustain its life-cycle



DEVELOPMENT, moulting and host-seeking (in moist leaf litter)

January March May July September November

FIGURE 59. Life-cycle of *Ixodes holocyclus* (paralysis tick) in relation to season, and to bandicoots and other hosts (redrawn from artwork by Stan Fiske, CSIRO Long Pocket Laboratories, see Sutherland & Tibballs 2001, Fig. 21.2).

Life-cycle and seasonality

Ixodes holocyclus is a three-host tick (Ross 1924), as are all of the other species of *Ixodes* that have been studied. One study in southeastern Queensland, revealed one major generation of *I. holocyclus* each year (Fig. 60). Yet the

presence of all life-stages at most times of the year indicates overlapping minor cohorts as well (Doube 1979). Nymphs were most abundant in April to September whereas adult female ticks are most abundant in October, November and December (Doube 1979, Fig. 60). Cases of tick paralysis due to *I. holocyclus* peaked in September in Brisbane (Fig. 60) which is intriguing since adult female ticks rather than nymphs cause paralysis in dogs (Doube 1975e). Copulation occurs in the grassy-nests of the primary natural hosts, *Isoodon macrourus* and *Perameles nasuta*, not on the host. Thus, male ticks are rarely seen on hosts (Moorhouse 1966, Moorhouse & Heath 1975). Copulation takes about one hour. The male apparently cuts off, with his chelicerae, the top of his spermatophore and then empties the contents of the spermatophore into the genital aperture of the female (Moorhouse 1966). Larvae and nymphs take 4 to 6 days to feed to repletion (i.e. engorged, Doube 1979) whereas adult females take 1 to 3 weeks to feed to repletion. Adult females feed to repletion in two phases: phase 1, slow imbibing of blood for typically 1–4 days but up to 2 weeks; and phase 2, the "big sip", rapid imbibing for 24 hours. The weight of females may increase 10 fold by full repletion (Fig. 59). Males feed on the haemolymph of adult females, rather than on the blood of their mammalian hosts. That males prefer to feed on females that are at least partially engorged indicates that the males are taking some of the mammalian blood that the females have imbibed (Moorhouse 1966). Thus male *I. holocyclus* may be parasites of female *I. holocyclus*. The mouthparts of males are very different to the mouthparts of females (Figs 54 & 56). The smaller teeth on the hypostome of males are apparently adapted for feeding on females. Males leave permanent feeding scars on the ventral surface of females e.g. Figure 3 of Moorhouse (1966). An individual female may have up to 15 feeding scars; such a female had been parasitised by males 15 times but the mean number of feeding scars per females was 2.9 (Moorhouse & Heath 1975). Whether or not the feeding of males on females leads to transmission of *Rickettsia australis* and/or other pathogens from male to female ticks or vice versa, is not known.

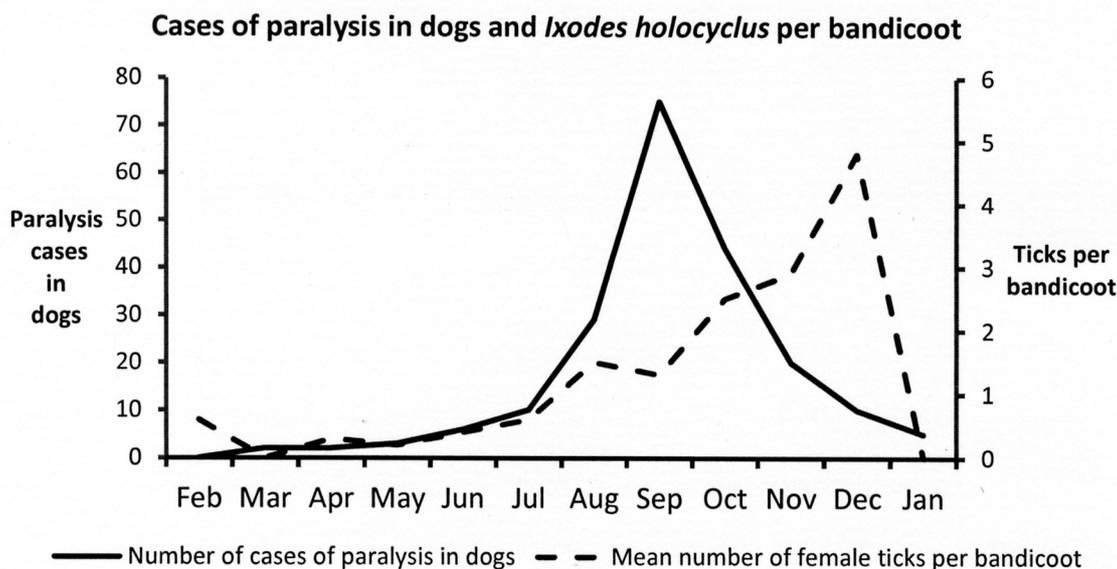


FIGURE 60. Numbers of cases of tick paralysis in dogs recorded at the Veterinary Clinic of The University of Queensland from 1970 to 1975 (solid line), and mean number of female *Ixodes holocyclus* (paralysis tick) per wild bandicoot found in southeastern Queensland from 1972 to 1974 (broken line) (data from Doube *et al.* 1977).

Although larvae, nymphs and adult female *I. holocyclus* will attach to, and attempt to feed on, almost any mammal and many species of birds, the bandicoots *Isoodon macrourus* and *Perameles nasuta* are needed to sustain populations of *I. holocyclus* in southeastern Queensland at least, from one year to another. For example, Doube (1979) found that many more larvae and nymphs fed to repletion on *I. macrourus* than on *Trichosurus vulpecula* (common brushtail possum), *T. caninus* (a short-eared brushtail possum), *Rattus rattus* (black rats) and *R. norvegicus* (brown rats). Thus, the presence of *I. holocyclus* in an area over several years indicates a stable population of *I. macrourus* and/or *P. nasuta* in that area.

Ixodes holocyclus does not produce cement when attaching to the skin of its host. It would be interesting to know whether or not the other six species in the subgenus *Sternalixodes* produce cement i.e. *I. confusus*, *I. cordifer*, *I. cornuatus*, *I. hirsti*, *I. myrmecobii* and *I. trichosuri*; probably they do not.

Disease

Paralysis of dogs, cats, livestock and wildlife. Paralysis caused by *I. holocyclus*, hereafter “tick paralysis”, is common in the regions where *I. holocyclus* lives. Dogs, cats, sheep and certain species of wildlife are particularly susceptible but calves and foals (Bootes 1962) are also susceptible. The father of Australian parasitology, Joseph Bancroft, was the first European to describe tick paralysis in Australia. Bancroft's description of tick paralysis in and around Brisbane was extraordinary in its clarity and insight (Bancroft 1884) especially since *I. holocyclus* had not yet been studied before nor even recognised as a species; that came in 1899 (Neumann, 1899). Bancroft described the pathology of tick paralysis in dogs and cats, and made the link between bandicoots and tick paralysis. What is more, he observed that “old dogs endure much longer than pups, and if the tick has been removed early recovery may be hoped for”.

Tick paralysis typically presents as a rapid ascending flaccid paralysis. Death usually results from paralysis of respiratory muscles. Other clinical signs that may be seen include a change in voice, vomiting, inappetance and anisocoria (pupils of unequal size) (Stone *et al.* 1989, Eppleston *et al.* 2013). Clinical signs of tick paralysis are not apparent until three days after *I. holocyclus* start to feed (Goodrich & Murray *et al.* 1978, Eppleston *et al.* 2013). Veterinarians often report variation in the intensity of the different clinical signs of tick paralysis in populations of dogs, from one year (tick season) to another. This is intriguing. For us, the variation is intelligible when tick paralysis is viewed as a multi-factorial disease. The interaction of at least three factors seem to determine the intensity of the different clinical signs from one year to another. First is the degree of innate immunity of the population of dogs. It is generally agreed that different breeds of dogs vary in susceptibility to tick paralysis (Seddon 1968) but this has not been studied quantitatively. Similarly, within a breed of dogs some populations and individual dogs seem to be less susceptible than others. It is likely that there has been both strong natural (Darwinian) selection and selection by human dog-breeders for genotypes of dogs less susceptible to tick paralysis. Second, the degree of acquired immunity of populations and indeed individual dogs may vary from one year to another. In the laboratory, dogs may acquire immunity (be immunised) if a single female is applied to a dog each week for nine weeks and the ticks are allowed to feed for increasing long periods until the dog is able to withstand full engorgement; for example, using the following regime: (i) allowed to feed to half-engorgement for the first three weeks; (ii) then to three-quarters engorgement for two weeks; (iii) then to full engorgement for a further two weeks; and (iv) then for a further two weeks two ticks rather than one are applied and are allowed to feed to engorgement (total period nine weeks) (Oxer 1948, Seddon 1968). If this dosing schedule is interrupted the dog will lose the acquired immunity (Oxer 1948, Seddon 1968). We envisage much variation in the “dosing schedule” of infection in the field and thus the degree of acquired immunity from one year to another. This is because the number of ticks that infect dogs varies from one year to another. Populations of ticks vary with, for example, summer rainfall, and the expertise and vigilance of tick-searching of dogs by dog-owners. The molecular composition of the toxin/s (eg Steen *et al.* 2006) might even vary from one year to another. This variation might be due to genetic variation among individual ticks, and perhaps populations, of ticks, and variation in epigenetic and other non-genetic factors that affect the final three dimensional structure of the toxin/s. Taken together, and considering that these three factors (innate immunity, acquired immunity and variation in the tick and its toxin/s) may interact, it is little wonder that the intensity of the different clinical signs of tick paralysis vary from one year to another.

Why might *I. holocyclus* cause paralysis, and often death, in its host? This question has long been debated e.g. Stone *et al.* (1989). The consensus view is that paralysis is an unintended side-effect of tick-feeding. Certainly, there is no obvious advantage to *I. holocyclus* to kill its host while feeding, whether that host be its preferred native host, a bandicoot, or a dog, cat, livestock species or a human. Paralysis is caused by female ticks; never male ticks. Nymphs may cause tick paralysis but this is usually clinically mild. Larvae may cause paralysis in cats but not dogs. Male ticks do not attach to nor feed on their hosts. Accordingly, the mouth parts of male *I. holocyclus* are very short (Fig. 57). In contrast, the mouth parts of female *I. holocyclus* are long (Fig. 55); the hypostome may perforate as deep as 1.7 mm (Allen *et al.* 1977).

Ixodes holocyclus are singular among the well-studied Australian ticks in that they do not produce cement at the bite site (Moorhouse 1969a). *Ixodes holocyclus* may cause paralysis and death in native eutherian (placental) mammals as well as in domestic eutherian mammals like dogs, cats, livestock and humans.

Since the late 1980s, *I. holocyclus* paralysis has killed many *Pteropus conspicillatus*, spectacled flying-foxes (bats), in North Queensland (e.g. Buettner *et al.* 2013), which is intriguing since *P. conspicillatus* lives high in sclerophyll and

rainforest trees. The current hypothesis is that *I. holocyclus* infest *P. conspicillatus* when *P. conspicillatus* feeds on the fruit of *Solanum mauritanum*, wild tobacco, an introduced plant that is now a weed in North Queensland (Eggert 1994, Buettner *et al.* 2013). Hall-Mendelin *et al.* (2011) and Stone (1988) reviewed *I. holocyclus* paralysis in the context of paralysis caused by other species of ticks in the Northern Hemisphere and Africa.

Alas, the number of dogs, cats, and livestock affected adversely by *I. holocyclus* has not been documented, but this would be most worthwhile. Stone (1988), however, speculated that 100,000 or more dogs, cats and livestock may be affected adversely by *I. holocyclus* to some degree in Australia each year, and that in New South Wales alone 10,000 cases of tick paralysis of dogs and cats were referred to veterinarians for treatment each year. These numbers seem overly large to us but the point is that the economic and social cost of *I. holocyclus* paralysis has not been adequately studied nor documented.

We highlight some other papers on tick paralysis in dogs, cats, livestock and wildlife. Webster *et al.* (2013a) reported on the histopathological changes in the lungs of 25 dogs that had tick paralysis. Webster *et al.* (2013b) reported the indications, durations and outcomes of mechanical ventilation in dogs and cats with *I. holocyclus* paralysis. Atwell (2011a) argued that *I. holocyclus* paralysis is not a truly ascending paralysis and that the larynx of dogs was invariably affected before the back legs, and thus change in the phonation, the dog's bark, was usually the first sign of *I. holocyclus* paralysis. Atwell (2011b) hypothesised "protective exposure" in dogs that had previously been infected with *I. holocyclus* compared to dogs that had not previously been infected with this tick. Large, long-term trials of dogs from tick-infested and tick-free areas are required, with well-defined exposure to *I. holocyclus* (Atwell, 2011b) will doubtless give new insight into the immune response of dogs to *I. holocyclus*. Bolton *et al.* (2011) described changes in white blood cell counts in dogs after *I. holocyclus* started to feed. Campbell & Atwell (2003) demonstrated acute left-sided heart failure in dogs with *I. holocyclus* paralysis. Atwell *et al.* (2001) proposed that therapy for dogs with tick paralysis should take into account the cardiopulmonary dysfunction not just the respiratory compromise, caused by infection with *I. holocyclus*. Doube *et al.* (1977) showed that two female *I. holocyclus* did not cause paralysis in two to three week old calves whereas four to 10 female *I. holocyclus* caused paralysis in all but one of 18 similar calves. Tick paralysis in cattle usually only occurs in young livestock, especially calves, although occasionally older animals succumb but usually when brought from an *I. holocyclus* free area to a ticky area (Doube 1975e). Twenty to 25 female *I. holocyclus* paralysed weaner-steers that weighed 80–160 kg (Doube 1975e), yet calves were protected if previously challenged with *I. holocyclus* (Doube & Kemp 1975 cited by Doube 1975e). This protection was due, at least in part to the lack of feeding, or slow feeding of *I. holocyclus* females on the previously challenged calves. Most of these ticks did not finish feeding and died *in situ*, attached to the calves (Doube & Kemp 1975).

Thurn *et al.* (1992) isolated a putative neurotoxin, "holocyclotoxin", from the salivary glands of *I. holocyclus*. This toxin (protein) may well be the toxin that causes tick paralysis in Australia or at least one of the toxins that causes tick paralysis in this country. Bootes (1962) reported fatal tick paralysis in five foals. Tick paralysis in adult horses has apparently been recorded only once in the scientific literature but this was in an adult miniature horse (Tee & Feary 2012). Clark & Clark (1969) reported a probable case of tick paralysis in *Isodon macrourus*; it was not clear if the bandicoot recovered or died.

Marks & Cribb (1966) reported tick paralysis in *Cracticus nigrogularis* (pied butcher bird) and a *Cracticus tibicen* (Australian magpie). The Australian magpie recovered but the pied butcher bird died, almost certainly from paralysis induced by a single *I. holocyclus* nymph or female. Domrow & Derrick (1965) reported the death of a *Platycercus elegans* (crimson rosella) from a single engorged female tick. *Ixodes holocyclus* may attack domestic fowl but tick paralysis due to *I. holocyclus* has not been reported in fowl (Seddon 1968).

Cats are usually meticulous groomers and thus find and remove ticks that a dog would not find and remove. Nonetheless, *I. holocyclus* may attach and stay attached in those areas that are difficult to groom, for example, between the shoulder blades and on the head and neck of a cat (Seddon 1968). Ticks may also be found on the flanks, the tail, behind the elbows and on the anus of cats (Seddon 1968). Cats are apparently less-susceptible, inherently, to tick paralysis compared to dogs (Seddon 1968, Arundel 1984).

Larval *I. holocyclus* may also cause tick paralysis, for example in cats (Fitzgerald 2007), and in *Mus domesticus* (laboratory mice) (Koch 1967). But this is rare and only occurs with large numbers of larvae. For example, at least 50 larval *I. holocyclus* were needed to kill a laboratory mouse whereas only five nymphs killed a similar sized mouse (Murray unpublished data cited by Koch 1967), and the case of paralysis in a cat involved 200–300 larval *I. holocyclus* (Fitzgerald 2007).

Paralysis and hypersensitivity from *I. holocyclus* in humans. Larval *I. holocyclus* often attach to humans but invariably the larvae are not able to feed and thus cause little more than localised dermatitis. In stark contrast to larvae, adult female *I. holocyclus*, and to a much lesser extent nymphs, often attach to humans and then over several days are able to feed and even engorge to repletion. Adult female *I. holocyclus* secrete a great range of chemical compounds into their hosts when feeding, especially after about day three of attachment. Some of these compounds may cause acute anaphylactic shock and paralysis. Medical staff who work along the east coast of Australia, where *I. holocyclus* lives, might have copies of Grattan-Smith *et al.* (1997) and Sutherland & Tibballs (2001) *et al.* close at hand. Sutherland and Tibballs (2001) have summaries of six instructive cases and guidance on first-aid and treatment of tick paralysis and a list of the 20 known fatalities caused by *I. holocyclus*. Considering the number of *I. holocyclus* in bushland along the east coast of Australia, the proclivity of *I. holocyclus* to attach to humans (and any other mammal) and thus the number of humans bitten each year, tick paralysis is a rare but potentially fatal, condition. Grattan-Smith *et al.* (1997) has summaries of six instructive cases of tick paralysis in children and guidance on clinical features and treatment. Grattan-Smith *et al.* (1997) was written by six medical graduates and a medical entomologist who work in Sydney. The following text is largely a précis of Sutherland and Tibballs (2001) and Grattan-Smith *et al.* (1997) with some ideas from us.

Paralysis is usually caused by adult female ticks; one tick is sufficient. Paralysis is most common in spring and summer but may occur at any time. Children 1–5 years of age are most commonly affected. The tick is usually found on the scalp; ticks attached to other parts of the body are less likely to cause paralysis. The typical presentation is a prodrome followed by the development of an unsteady gait, and then ascending symmetrical, flaccid paralysis. In children, usually the child becomes subdued and refuses food. Sometimes excessive periods of sleep occur and the child is difficult to wake and is found to be weak and obviously ill when roused for feeding or bathing. Over the next 24 hours ascending, symmetrical weakness and paralysis may progress to involve the upper limbs and terminally; the muscles involved in swallowing and breathing become paralysed. In older children and adults, the initial complaint may be difficulty in reading with double vision, photophobia, nystagmus or papillary dilation. Usually the body temperature is normal or subnormal unless the tick infestation is complicated by a bacterial infection. Early cranial nerve involvement is a feature, particularly the presence of both internal and external ophthalmoplegia. Paralysis may worsen 24 to 48 hours after the tick is removed, thus the child must be observed carefully during this period. Death from respiratory failure, as in *I. holocyclus* paralysis of dogs, was relatively common in the first half of the twentieth century. Intensive care units at hospitals and medical knowledge have made tick paralysis far less common in Australia. Indeed, there has not been a fatality since 1945. Yet tick paralysis is still potentially fatal. Wherever possible, cases of tick paralysis, especially in children, should be managed in intensive care units. Respiratory support may be required for over a week, but full recovery occurs. This is slow with several weeks passing before the child can walk unaided. The first step in treatment is to find the tick and then remove it. Hamilton (1940) stressed that more than one tick may be attached and that a careful search of body crevices should be made, including the auditory meatus, natal cleft, the vulva and nose. It is common, perhaps usual for the attached tick to be misidentified as an innocuous swelling of the skin, by patients, family members and even medical staff. This is because the tick is invariably darkish from the blood-meal and thus blends in with all but blond hair and very pale skin, and the recently attached or partially engorged tick looks and feels much like a soft outgrowth of the skin. The mouth parts are invariably buried deep in the skin so all that is obvious is the soft main body.

Ticks should be removed by grasping the tick either side of the tick's mouth parts, pressing well down into the skin, avoiding pressure on the main body of the tick. The tick can then be levered straight out with steady tension. The anti-tick paralysis serum, which is raised in hyper-immune dogs, has a role in treatment of seriously ill children but there is a high incidence of acute allergy and serum sickness. Ticks should not be removed with the fingers since some experts claim that this is likely to push some of the gut contents of the tick into the wound and thus potentially worsen the condition. This claim has not been proven, however. Stone (1988, 1990) recommended trying to kill the tick *in situ* with insecticide sprays but in the experience of the present authors, the longer the tick is attached the sicker the person becomes, at least in adults. Inokuma *et al.* (2003) presented an interesting case report of a 59 year old Japanese tourist to the Gold Coast, Queensland, to whom a female *I. holocyclus* attached. The semi-engorged tick was discovered three days later by himself, in his homeland, Japan. Tick paralysis may be mistaken for poliomyelitis, myasthenia, botulism, myopathies and a variety of inflammatory and toxic neuropathies. On the east coast of Australia, tick paralysis should be considered in all putative cases of Guillain-Barré syndrome, particularly if there is early pupillary involvement.

Larvae, nymphs and adult *I. holocyclus* may sensitise a patient and precipitate an allergic reaction, ranging from rapid and gross swelling to life-threatening anaphylactic shock. Sutherland & Tibballs (2001) advise that mild allergic reactions may resolve after the tick is found and removed, and antihistamines have been administered. In severe cases, adrenalin may be required as well as measures to ensure adequate ventilation. Usually there is a clear history of worsening reactions, and the patient should: i) carefully avoid contact with ticks; ii) have access to injectable adrenalin at all times, and iii) if so advised by a specialist, receive immunotherapy with tick extracts to reduce the state of the allergy.

Treatment of tick paralysis. Anti-tick paralysis serum, raised in hyper-immune dogs, is available in Australia to treat both dogs, and humans (see above). The dog antiserum has been used to treat, successfully, cats, calves, foals other mammals and even birds.

Pathogens transmitted to humans by *I. holocyclus*. *Rickettsia australis*, a bacterium which causes Queensland tick typhus, is transmitted to humans by *I. holocyclus*. *Rickettsia australis* is one of two spotted fever group *Rickettsia* spp. known from Australia: the other is *R. honei* (Flinders Island spotted fever) which is transmitted to humans by *Bothriocroton hydrosauri*. Although Queensland tick typhus is usually a mild disease, there was a serious illness and death in Mossman, North Queensland (Sexton *et al.* 1990). Symptoms of Queensland tick typhus include fever, headache, malaise, enlarged lymph nodes and a macropapular (maybe vesicular) rash. Most patients have an eschar and some have a slight cough, myalgia and chills (Brody 1946, Andrew *et al.* 1946). Playford & Whitby (1996) provided photographs of the eschar and rash on a patient with Queensland tick typhus. Queensland tick typhus is only known, so far, from the eastern coastal areas of Australia from Darnley Island in the Torres Strait in the north (Unsworth *et al.* 2007a) to Wilson's Promontory in Victoria in the south (Graves & Stenos 2009), which, intriguingly, is outside of the known geographic distribution of *I. holocyclus* (Fig. 58). The presence of Queensland tick typhus in the Torres Strait is also intriguing since the vector of *R. australis*, *I. holocyclus*, has not yet been collected north of Shipton's Flat, 35 km south of Cooktown, Queensland (see section entitled Habitat & Geographic Range). Collections of *I. holocyclus* north of Shipton's Flat would be worthy of a note in the scientific literature. Wilson *et al.* (2013) reported three cases of Queensland tick typhus with unusual complications: splenic infarction, myocarditis and leukocytoclastic vasculitis, in addition to the usual manifestations.

Queensland tick typhus was known as North Queensland tick typhus from 1946 when it has first recognised as a clinical entity, in Mossman, North Queensland (Brody 1946, Andrew *et al.* 1946) until the late 1980s (e.g. Stone 1988 p 65). Now this disease is known as Queensland tick typhus, despite the fact that its geographic range extends to Wilson's Promontory, the most southerly point of mainland Australia! The genome of *R. australis* has been sequenced (Dong *et al.* 2012).

Erythema migrans (literally "migrating redness", due to dilation of blood capillaries) at and around the site on the skin where *I. holocyclus* attaches is not uncommon (see Figs 1 & 2 of Mayne 2012 for photographs of erythema migrans at the bite site of ticks, probably *I. holocyclus*). Experience from other countries shows that erythema migrans at the site of attachment of ticks may be due to a range of different bacteria including *Rickettsia* spp., *Bartonella* spp., *Coxiella burnetii* (Q fever), *Spiroplasma* spp. and *Borrelia* spp. Whether or not a species of *Borrelia* is transmitted to humans by *I. holocyclus* has been controversial for at least 20 years (Russell 1995; Russell *et al.* 1993, 1994; Mayne 2011, 2012) and will remain controversial until a *Borrelia* organism is isolated from a patient who was bitten by *I. holocyclus* and has symptoms of borreliosis. Lyme borreliosis is a disease caused by *Borrelia burgdorferi* transmitted by *Ixodes* spp. in the Northern Hemisphere. *Borrelia burgdorferi* has not been found in Australian wildlife, ticks or humans who have not recently travelled to endemic areas of Lyme disease. Yet it is possible that an endemic Australian *Borrelia* species may be transmitted to humans, from time to time, in Australia, by the bite of *I. holocyclus*.

Barker (2003) hypothesised that *I. holocyclus* may be a link in the transmission of Hendra virus from bats to horses to humans; this hypothesis has not yet been tested.

Habitat and geographic range

Ixodes holocyclus may be found in habitats ranging from dry sclerophyll forest with a thick understory of grass (e.g. at The University of Queensland Veterinary Research farm at Pinjarra Hills, Brisbane) to wet sclerophyll forest with a thick understory of shrubs (e.g. Mt Tamborine area, west of the Gold Coast, Queensland) to rainforest (e.g. Milla Milla, North Queensland). Rainfall and thus humidity may be good predictor of the presence/absence of

I. holocyclus (Heath 1974). In Queensland, *I. holocyclus* is said to be found in areas that have at least 1,000 mm rainfall per year (Heath 1974). The susceptibility of eggs and larvae to desiccation in low humidity environments in summer is apparently one of the main factors behind the geographic distribution of *I. holocyclus* (Shaw 2000). The other main factor is that stable populations of bandicoots, *Isodon macrourus* and/or *Perameles nasuta* are needed for population of *I. holocyclus* to persist. The absence of *I. holocyclus* from Cape York Peninsula requires explanation since intuitively and empirically (Heath 1974) the climate and vegetation of Cape York Peninsula seems to be suitable for *I. holocyclus* (Shaw 1997). The absence, or at least scarcity, of *I. macrourus* and *P. nasuta* on Cape York Peninsula is the most compelling hypothesis for the absence of *I. holocyclus* from Cape York Peninsula (Shaw 1997). The maps in Dickman & Stodart (2008) and Gordon (2008) indicate that *I. macrourus* and *P. nasuta* occur on Cape York Peninsula but these maps show the known *historical* distribution of these bandicoots since *P. nasuta* was last recorded on Cape York Peninsula in 1948 and is now either scarce (Winter & Allison 1988, Gordon *et al.* 1990) or extinct in this region. Similarly, we cannot find recent evidence of *I. macrourus* on Cape York Peninsula. The most northerly confirmed locality of *I. holocyclus* is Shipton's Flat, 35 km south of Cooktown, Queensland (Song *et al.* 2011). Unsworth *et al.* (2007a) found Queensland tick typhus on Darnley Island, Torres Strait, Queensland so *I. holocyclus*, the main vector of this bacterium, might conceivably be on Darnley Island. Banyard (2013) reported a single female *I. holocyclus* from Namadgi National Park, ACT. This record needs to be confirmed with additional collections in the ACT.

Genes and genomes

Parts of the following genes are available to the public in GenBank: (i) small subunit nuclear rRNA (18S) (1747 bp, Dobson & Barker 1999); (ii) Internal Transcribed Spacer (ITS2) rRNA (568 bp, Song *et al.* 2011; see also Hlinka *et al.* 2002 for a secondary structure of ITS2, and Shaw *et al.* 2002); (iii) *cytochrome c oxidase subunit I* (*COX I*) (644 bp, Song *et al.* 2011); (iv) entire mitochondrial genome (15,007 bp Shao *et al.* 2005a); (v) holocyclotoxin-1 (222 bp, Thurn *et al.* 1992); (vi) 28S ribosomal RNA gene (644 bp, Klompen *et al.* 2000); and (vii) 5.8s ribosomal RNA gene (ca 150 bp, Hlinka *et al.* 2002).

The mitochondrial genome of *I. holocyclus* and the 27 other species in the Australasian *Ixodes* lineage are notable since these genomes have duplicate non-coding regions (Shao *et al.* 2005a, Fig. 2 Shao & Barker 2007).

Other information

Ixodes holocyclus and *I. cornuatus* are the two main causes of tick paralysis in mammals, mainly in dogs and cats, in Australia, but Kemp (1979) proposed that all seven species in the subgenus *Sternalixodes* may cause paralysis in their hosts: *I. holocyclus*, *I. cornuatus*, *I. confusus*, *I. cordifer*, *I. hirsti*, *I. myrmecobii*, and *I. trichosuri*. Future workers might seek to confirm this idea; there are published records of tick paralysis for only *I. holocyclus*, *I. cornuatus* and *I. hirsti* (Kemp 1979). Murrell *et al.* (2003) cultured bacteria from *I. holocyclus* and some other Australian ticks.

Ixodes tasmani Neumann, 1899 (common marsupial tick)

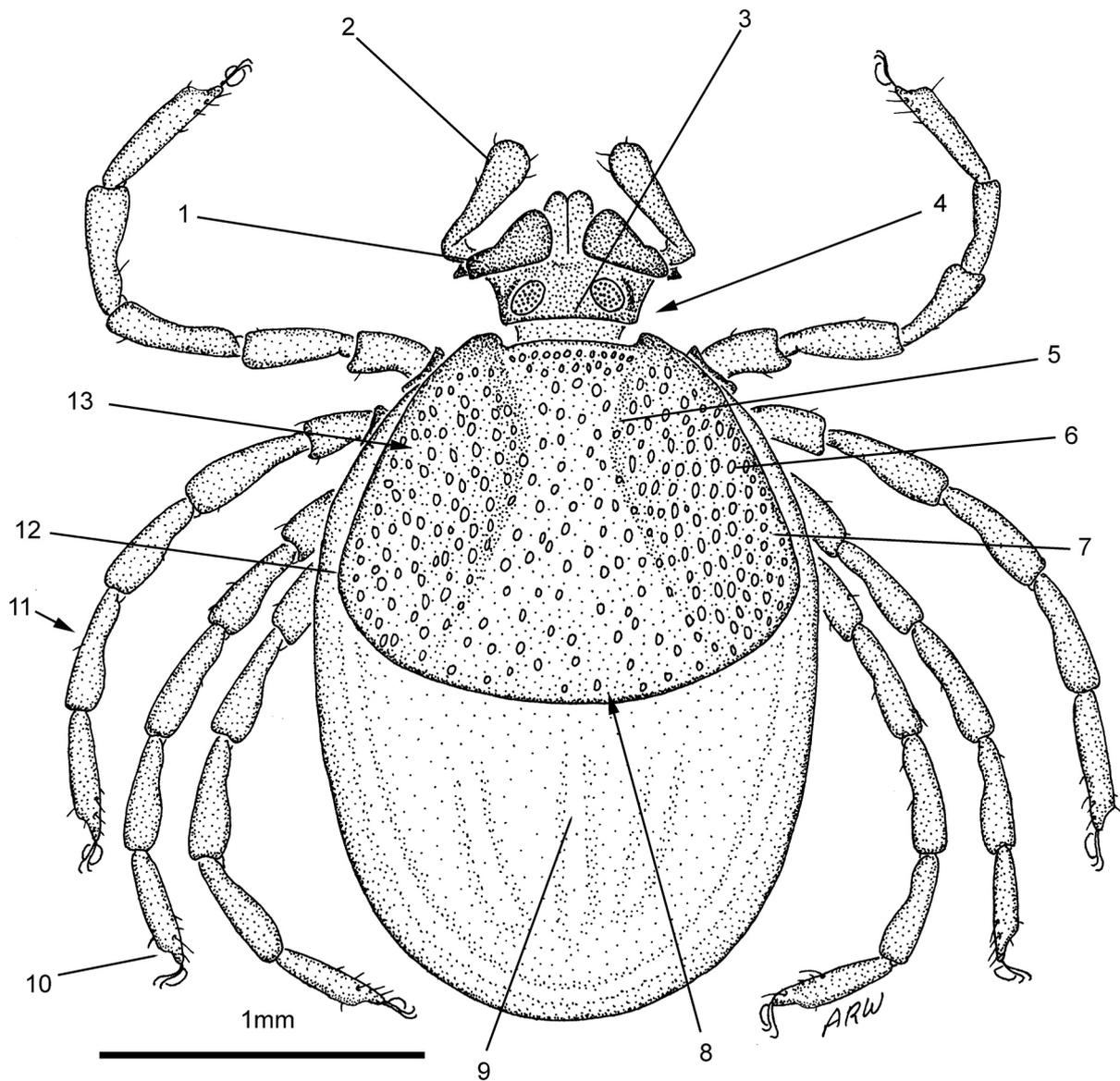
General

Ixodes tasmani is known as the common marsupial tick on account of its extraordinarily catholic feeding habits and widespread geographic distribution in Australia. *Ixodes tasmani* was named after the explorer and mariner Abel Tasman. *Ixodes tasmani* is one of 28 species in the Australasian lineage of *Ixodes* (Barker & Murrell 2002, 2004) and one of seven species in the subgenus *Exopalpiger* in Australia.

Differential diagnosis

The following refers only to adult females (Figs 61 & 62). *Ixodes tasmani* is distinctively different from the other three *Ixodes* species in this book. It has mouthparts with widely diverging palps, with the first palpal article (segment) aligned so that the palps overlap the cheliceral sheaths whilst pushing the outer palpal articles away from the cheliceral sheaths. Also the piercing mouthparts (chelicerae and hypostome) of *I. tasmani* are short compared to the other *Ixodes* species in this book. Of the four *Ixodes* species in this book, *I. tasmani* is the only species with a scutum bearing dense large punctations. In specimens that are not too expanded by feeding, the anal groove can be

seen as an open inverted U-shape, compared to the closed pointed shape of the posterior part of the anal groove of the other species.



1 Palp article 1 is widely angular and extends over cheliceral sheath.

2 Palps shape is short and thick.

3 Basis dorsal posterior margin is straight.

4 Cornua are absent.

5 Cervical grooves are long.

6 Punctuation size on scutum is large.

7 Punctuation distribution on scutum is dense.

8 Scutal corrugations at posterior are absent.

9 Posterior grooves are present.

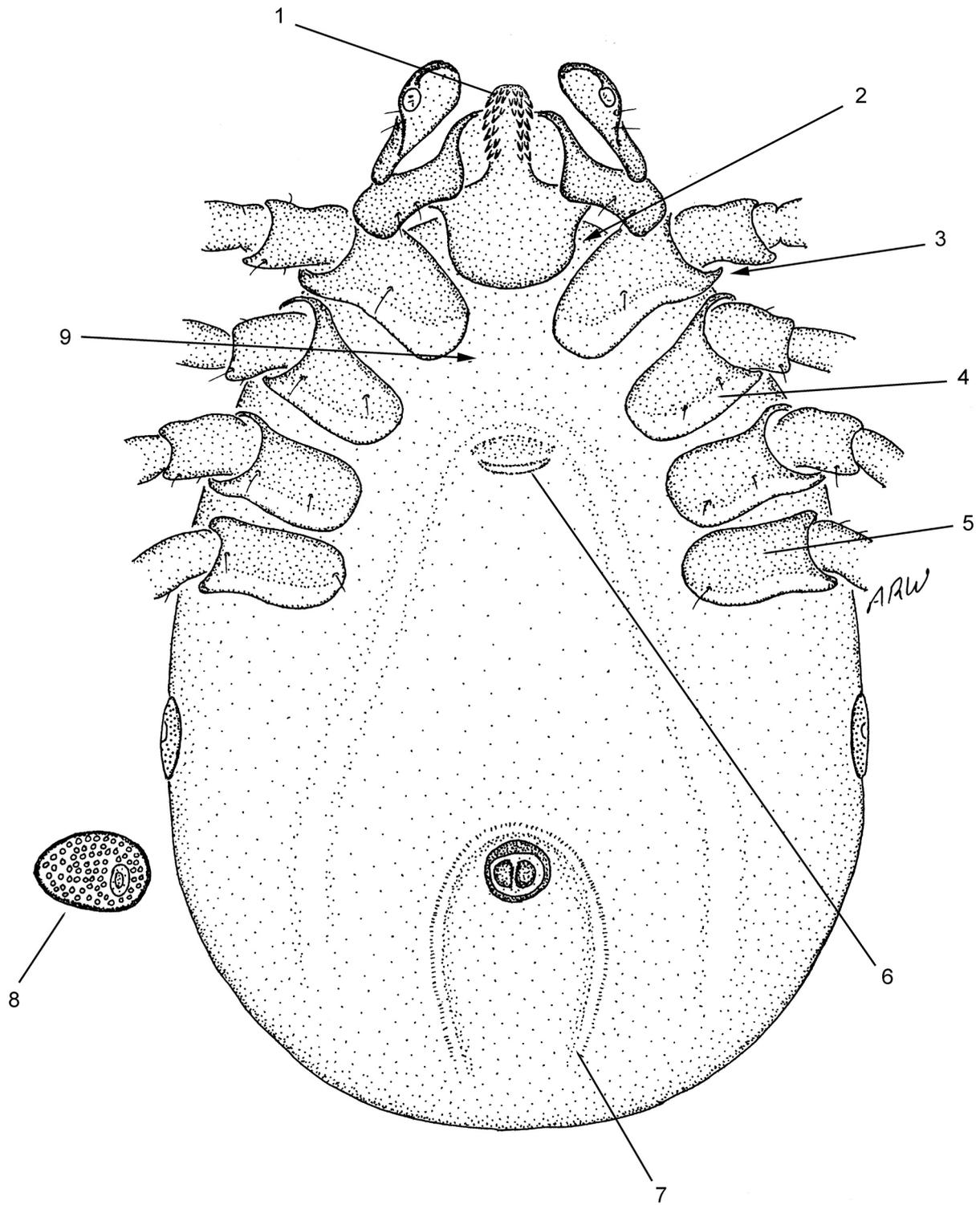
10 Tarsi distal profile is steeply stepped.

11 Leg colour pattern is absent.

12 Scutal proportion is wider than long.

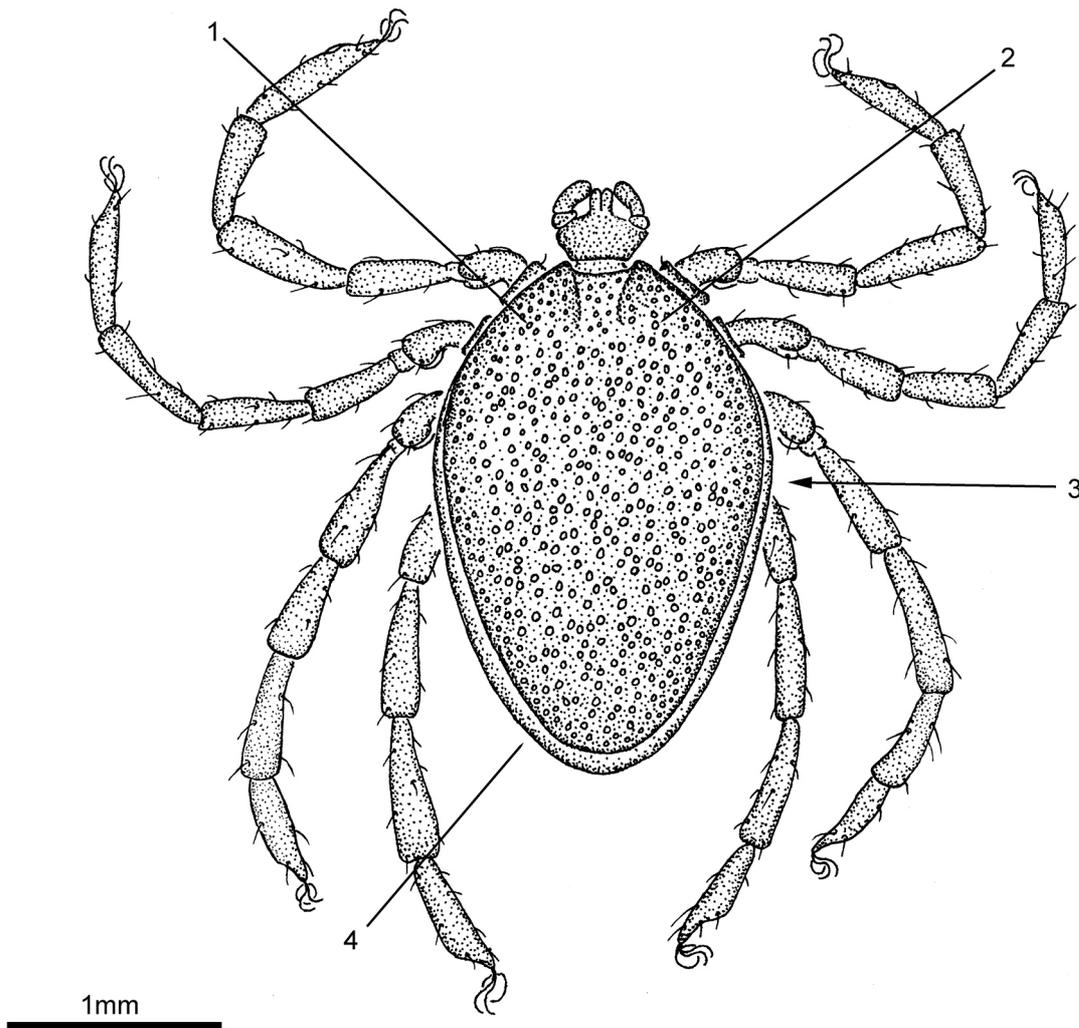
13 Lateral carinae are absent.

FIGURE 61. *Ixodes tasmani* (common marsupial tick), female, dorsal.



- | | |
|--------------------------------------|---|
| 1 Hypostome shape widens anteriorly. | 6 Genital aperture position is level with coxae 2 to 3. |
| 2 Auriculae are absent. | 7 Anal groove shape is open at posterior. |
| 3 Coxae 1 external spur is absent. | 8 Spiracle plate shape is oval. |
| 4 Coxae type is with syncoxae. | 9 Sternal plate is absent. |
| 5 Coxae surface texture is smooth. | |

FIGURE 62. *Ixodes tasmani* (common marsupial tick), female, ventral.



- 1 Punctation size on conscutum is large.
- 2 Punctation distribution on conscutum is dense.
- 3 Lateral groove on conscutum is absent.
- 4 Body profile is a narrow oval.

FIGURE 63. *Ixodes tasmani* (common marsupial tick), male, dorsal.

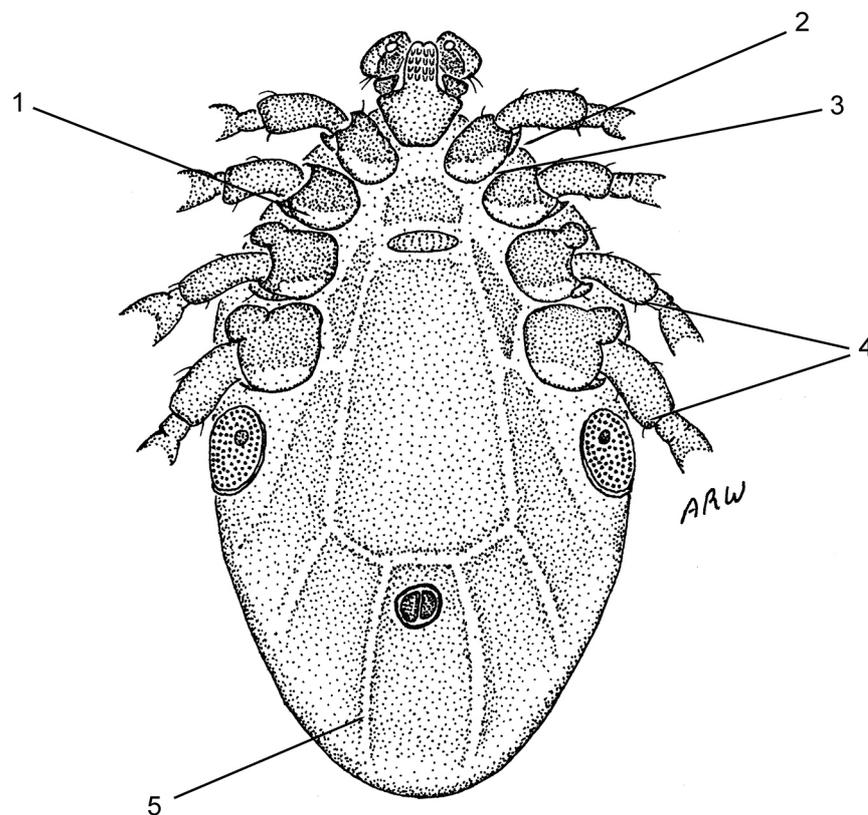
Hosts

Host records from Roberts (1970): *Tachyglossus aculeatus* (short-beaked echidna), *Antechinus flavipes* (yellow-footed antechinus), *A. swainsonii swainsonii* (Tasmanian dusky antechinus), *A. s. mimetes* (dusky antechinus), *Phascogale tapoatafa* (brush-tailed phascogale), *Sminthopsis murina* (common dunnart), *Perameles gunnii* (eastern barred bandicoot), *P. nasuta* (long-nosed bandicoot), *Isoodon obesulus* (southern brown bandicoot), *Trichosurus caninus* (short-eared brushtail possum), *T. vulpecula vulpecula* (common brushtail possum), *T. v. fuliginosus* (Tasmanian brushtail possum), *Gymnobelideus leadbeateri* (Leadbeater's possum), *Petaurus australis* (yellow-bellied glider), *P. breviceps* (sugar glider), *Pseudochirops archeri* (green ringtail possum), *P. peregrinus convolutor* (common ringtail possum), *P. p. peregrinus* (common ringtail possum), *Petauroides volans* (greater glider), *Phascolarctos cinereus* (koala), *Cercartetus lepidus* (little pygmy-possum), *Vombatus ursinus* (common wombat), *Potorous tridactylus* (long-nosed potoroo), *Bettongia gaimardi* (Tasmanian bettong), *Petrogale lateralis pearsoni* (Pearson Island rock-wallaby), *Thylogale billardierii* (Tasmanian pademelon), *T. stigmatica* (red-legged pademelon), *Macropus rufogriseus* (red-necked wallaby), *Homo sapiens* (man), *Oryctolagus cuniculus* (rabbit, introduced), *Conilurus albipes* (white-footed rabbit-rat, extinct), *Pseudomys higginsii* (long-tailed mouse), *Mastacomys fuscus* (broad-toothed rat), *Melomys cervinipes* (fawn-footed melomys), *Uromys* sp., *Rattus sordidus*

(canefield rat), *R. fuscipes* (bush rat), *R. lutreolus* (swamp rat), *R. norvegicus* (brown rat, introduced), *R. rattus* (black rat, introduced), *Hydromys chrysogaster* (water rat), *Canis familiaris* (domestic dog, introduced), *Felis catus* (domestic cat, introduced) and *Equus caballus* (domestic horse, introduced).

Ixodes tasmani is catholic in its feeding habits, it has been recorded from 42 species of hosts and most families of Australian marsupials, monotremes, rodents, domestic animals and humans but not from birds nor reptiles. In comparison, *I. holocyclus* has been recorded from 36 species of mammals and seven species of birds (refer to the *I. holocyclus* species account).

Host records since Roberts (1970) (from three different sources): (i) *Antechinus agilis* (referred to as *A. stuartii*; Beveridge and Barker 1976); (ii) *Antechinus agilis* (agile antechinus), *R. fuscipes* (bush rat), *R. lutreolus* (swamp rat), *Sminthopsis leucopus* (white-footed dunnart) and *Isoodon obesulus* (southern brown bandicoot) (Weaver & Aberton 2004); and (iii) *Isoodon macrourus* (northern brown bandicoot), *I. obesulus* (southern brown bandicoot), *Perameles nasuta* (long-nosed bandicoot), and *Trichosurus caninus* (short-eared brushtail possum) (Barker & Campelo unpublished data).



- 1 Coxae type is with syncoxae.
- 2 Coxae external spurs are absent.
- 3 Coxae internal spurs are absent.
- 4 Trochanter spurs on legs 3 and 4 are indistinct.
- 5 Anal groove shape is not joined at posterior.

FIGURE 64. *Ixodes tasmani* (common marsupial tick), male, ventral.

Life-cycle and seasonality

Ixodes tasmani is a three-host tick (Murdoch & Spratt 2005). In the laboratory, the entire life-cycle was completed in four months. This observation, together with the presence throughout the year of all life-stages in central-eastern New South Wales (Murdoch & Spratt 2005) and in Brisbane, Queensland (Gemmell *et al.* 1991), led Murdoch & Spratt (2005) to conclude that there were multiple generations of *I. tasmani* per year. Female *I. tasmani* were attached to laboratory rats (*Rattus norvegicus*) for an average of four days which was similar to six days reported

by Heath (1986). Mating occurred in tubes; thus, in nature mating may occur off the host. The average time from when females engorge and detach to when they start laying eggs (the preoviposition period) was 14 ± 3 days. Eggs hatched on average 39 ± 8 days after being laid. Larvae feed successfully on laboratory rats as did nymphs. Larvae feed for two to three days whereas nymphs feed for one to five days (average 3.2 ± 0.9 days). Heath (1986) also found that larvae were attached for three days. The average premoult period (nymphs to adults) was 27 ± 3 days (Murdoch & Spratt 2005). Most larvae, nymphs and adults detached from their hosts during the "daylight" hours i.e. detachment was diurnal (Murdoch & Spratt, 2005). Detachment during the "daylight" hours would have these detached ticks in or near their hosts where their hosts were sleeping during the day. Murdoch & Spratt (2005) concluded that *I. tasmani* was a nidicolous tick i.e. a tick that lives in and around the resting place of its host e.g. the dens of the common brushtail possum (*Trichosurus vulpecula*) in hollow trees. Heath (1986) also studied seasonality in *I. tasmani* (in Queensland).

Adult and nymphal *I. tasmani* were found on *T. vulpecula* in central-eastern New South Wales throughout the year, yet larvae were found on this host only in summer and autumn (November to March, Fig. 3, Murdoch & Spratt 2005). Heath (1986) also found adult *I. tasmani* throughout the year.

Ixodes tasmani produces cement when attaching to its host whereas, curiously the other *Ixodes* species that has been studied, *I. holocyclus*, does not (Moorhouse 1969a). It would be interesting to know whether or not the other *Ixodes* species in Australia produce cement. *Ixodes tasmani* has short mouth parts for a female *Ixodes* tick (Fig. 62); cement is an adaptation to these short mouthparts.

Disease

Gemmell *et al.* (1991) showed that juvenile *Isoodon macrourus* (northern brown bandicoots) that were infected with *I. tasmani*, *I. holocyclus* and *Haemaphysalis humerosa* were anaemic, had elevated white blood cell counts and grow 28% slower (1.8 g versus 2.5 g per day) than tick-free bandicoots. These data lend weight to the widespread idea that juvenile bandicoots, and perhaps juveniles of other marsupial species, are especially vulnerable to infections and disease immediately after weaning (references in Gemmell *et al.* 1991) but it is not known whether *I. tasmani*, *I. holocyclus*, *H. humerosa* or the combined effect of these three species of ticks caused the slower weight gain and changes in haematology in the bandicoots.

Tick paralysis has not been associated with *I. tasmani* and domestic animals (Seddon 1968) nor wildlife but there have been few studies.

Since *I. tasmani* has the most catholic feeding habits of any Australian tick, this tick has been examined by PCR for potentially pathogenic bacteria. Five different types of bacteria, or more precisely DNA of five different types of bacteria, have been found in *I. tasmani* so far: a spotted fever group *Rickettsia* sp., *Rickettsia tasmanensis*, a *Rickettsiella* sp., a *Hepatozoon* sp. and a *Bartonella*-like sp. (Vilcins *et al.* 2008, 2009a, 2009b, 2009c; Izzard *et al.* 2009). Similar PCR studies of other ticks may well also reveal bacteria that are potentially pathogenic to wildlife, domesticated animals and humans. *Rickettsia australis*, the aetiological agent of Queensland tick typhus, has also been isolated from *I. tasmani* in southeast Queensland (Campbell & Domrow 1974). Weilgamma (1986) showed that *I. tasmani* can be a vector of *Theileria peramelis*.

Spratt and Haycock (1988) found third-stage larvae of the filarioid nematode *Cercophitifilaria johnstoni* in newly moulted nymphal and adult *I. tasmani* after these ticks had fed on *Rattus fuscipes* that were infected with this nematode (Spratt, pers. comm.). The discovery by Spratt & Haycock (1988) that *I. tasmani* become infected with the nematode *C. johnstoni* was the solution to a question posed by Moorhouse (1969b). Moorhouse was puzzled by the sight of microfilariae on the mouthparts and in the tissue under the skin where *I. tasmani* were attached to a marsupial glider, *Petauroides volans* (greater glider) (Moorhouse, 1969b). It seems that Moorhouse could not believe his eyes; that this tick was a putative vector of a filarioid nematode. Thus, Moorhouse did not hypothesize that *I. tasmani* was a vector of *C. johnstoni*, rather he hypothesized that the microfilariae were attracted to "some chemotactic substance present in the saliva" of the tick (Moorhouse 1969b).

Habitat and geographic distribution

Although *I. tasmani* is probably the most widespread of the Australian *Ixodes* species, there are only a handful of precise locality-records in the scientific literature. The following text is from Roberts (1970). "This tick is certainly the most common and abundant species in Tasmania and has been recorded there in numerous localities. It is also widespread in Victoria. In New South Wales, there are records of its occurrence throughout the entire coastal and

sub coastal areas with inland extensions to Moree, Dubbo, and Kosciusko. The species is known in Queensland throughout the coastal and sub coastal areas from Iron Range in the north and inland to Emerald and Roma. There are also several records from southeastern South Australia and south-western Western Australia."

Genes and genomes

Parts of the following genes are available to the public in GenBank: (i) large subunit nuclear rRNA (28S) (Klompen *et al.* 2000); (ii) small subunit nuclear rRNA (18S) (Klompen *et al.* 2000); (iii) large subunit rRNA (16S, mitochondrial) (Norris *et al.* 1999); and (iv) small subunit rRNA (12S, mitochondrial) (Norris *et al.* 1999).

Other information

None.

***Rhipicephalus (Boophilus) australis* Fuller, 1899 (Australian cattle tick)**

General

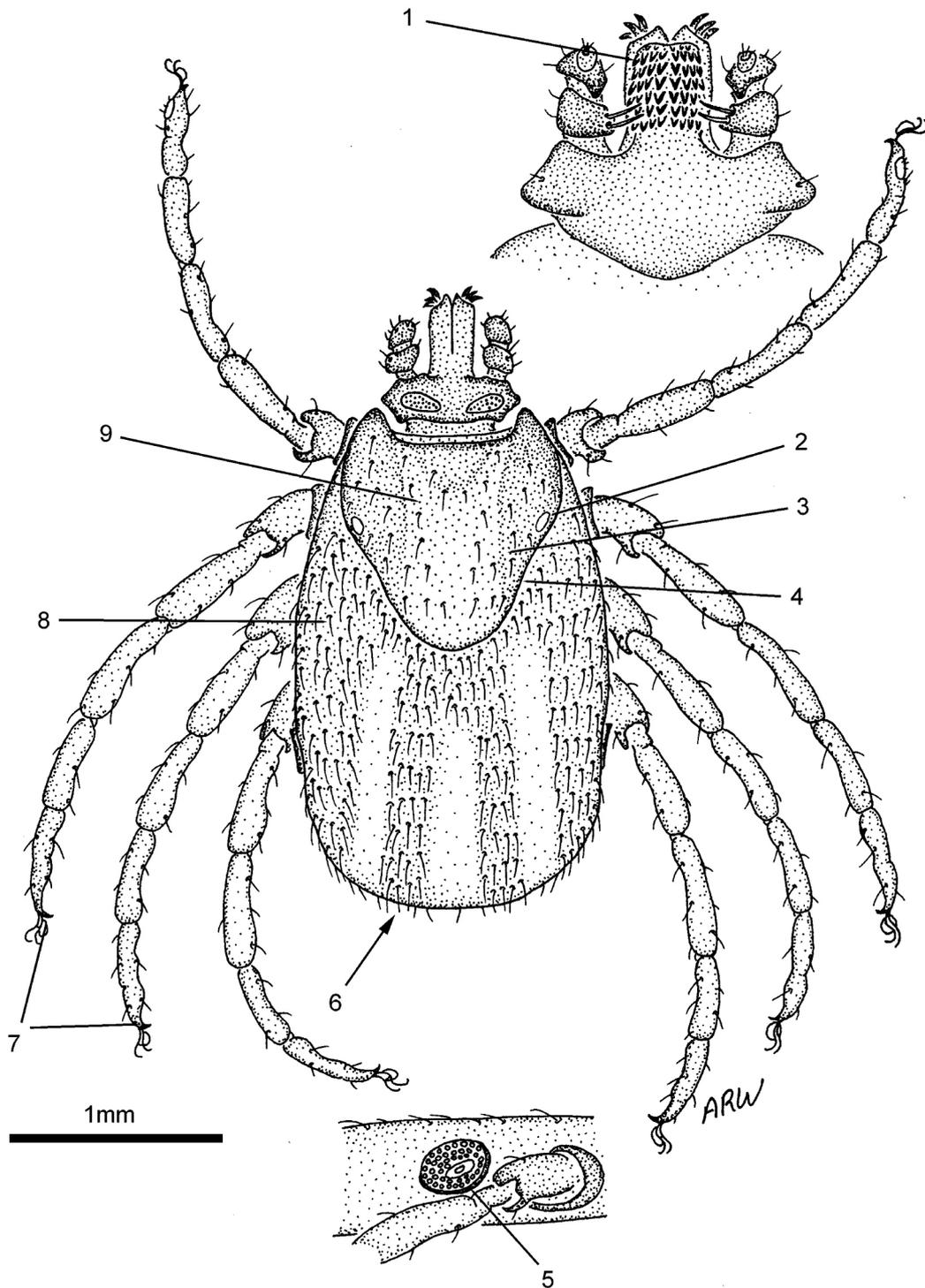
Rhipicephalus (Boophilus) australis is known as the cattle tick in Australia (Carne *et al.* 1987) but we propose the common name "Australian cattle tick" since its Linnean species name is now "*australis*". Note that the Australian cattle tick had the name *Boophilus microplus* until 2012 (Estrada-Peña *et al.* 2012); a full account of the taxonomy and names of this tick is given in "Other information", below. *Rhipicephalus australis* is one of at least four species in the *Rhipicephalus microplus* complex of species: (i) *R. australis*; (ii) *R. microplus*—Clade A species; (iii) *R. microplus*—Clade B species; and (iv) *R. annulatus* (Burger *et al.* 2014b). The practical implications of the realization that cattle ticks that were once known as *R. microplus* from different parts of the world comprise at least three different species are substantial. It can no longer be assumed that the results of experiments and field studies on cattle ticks from Australia (*R. australis*) are pertinent to cattle ticks from North America, South America, Africa and northern China (*R. microplus*—Clade A species), and vice versa, nor to southern China (*R. microplus*—Clade B species).

Differential diagnosis

In our hands, *R. australis* cannot be distinguished unambiguously from the two putative undescribed species of *R. microplus* (*R. microplus*—Clade A species and *R. microplus*—Clade B species) and at times, even *R. annulatus* (see also Burger *et al.* 2014b). Indeed, for us, it is a myth that *R. australis*, *R. microplus* and all *R. annulatus* can be distinguished unambiguously with morphology *without reference to geography*. The morphological characters are just too variable among populations of these ticks. Other authors have had similar experiences with *R. microplus*, *R. annulatus* and *R. decoloratus* e.g. Uilenberg (1962), and Lempereur *et al.* (2010). The species of the subgenus *Boophilus* are patently very closely related to one another and morphologically similar. Fortunately, it seems that species of the subgenus *Boophilus* are genetically distinguishable (Burger *et al.* 2014b and references therein) and that a 865 bp of *cytochrome c oxidase I (cox 1)* nucleotide sequence can distinguish, unambiguously, the seven putative species in the subgenus: *R. annulatus*, *R. australis*, *R. decoloratus*, *R. geigy*, *R. kohlsi*, *R. microplus*—Clade A species and *R. microplus*—Clade B species (Burger *et al.* 2014b). We caution against relying on morphology alone to identify species of the subgenus *Boophilus*, especially since it is not known how many species are in the subgenus and the geographical distributions of boophilid ticks are changing by the historically unprecedented movement in this century of live cattle and other hosts. Take for example, the apparently fluid geographic distribution of *R. microplus*, *R. decoloratus*, *R. geigy* and *R. annulatus* in west Africa (Lempereur *et al.* 2010).

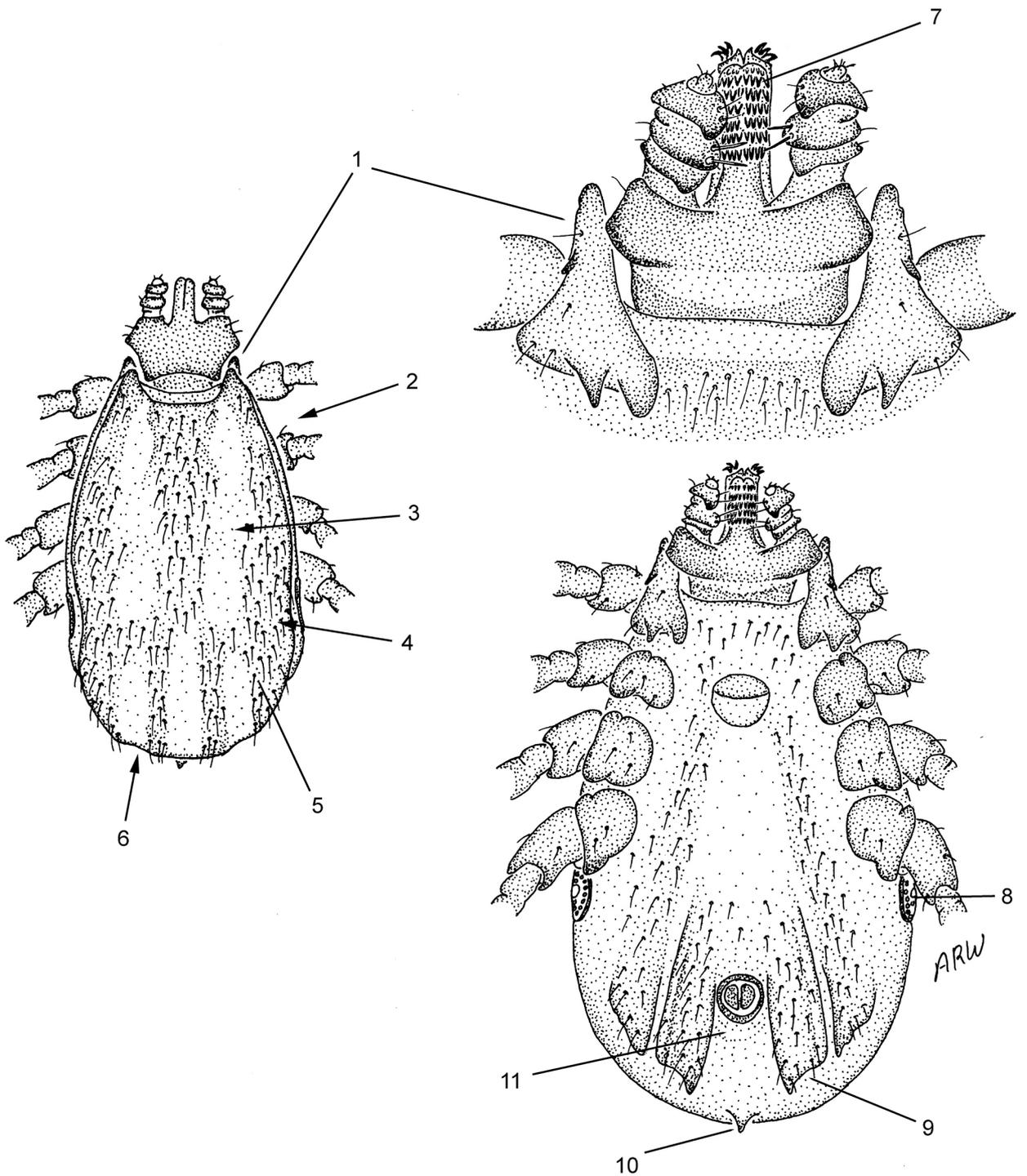
Using morphological characters, however, *R. australis* can be differentiated easily from *R. sanguineus*, the only other species of *Rhipicephalus* in Australia (Figs 65 & 66). Female *R. australis* from the dorsal surface has a scutum that has: shallow and indistinct cervical fields, a single concavity on each side of the posterior scutum, and eyes that are difficult to distinguish from the surrounding scutum. In contrast, *R. sanguineus* has a scutum that has: deep and distinct cervical fields, a double concavity on each side of the posterior scutum, and easily visible eyes (Figs 69 & 70). The setae on the scutum and alloscutum of *R. australis* are dense and long whereas those of *R. sanguineus* are sparse and short. On the lateral and ventral surfaces of females, *R. australis* has small circular

spiracle plates and 4 + 4 columns of teeth on the hypostome. In contrast, *R. sanguineus* has large comma-shaped spiracle plates and a hypostome with 3 + 3 columns of teeth.



- | | |
|---|---|
| 1 Columns of teeth on hypostome are 4 + 4. | 6 Festoons are absent. |
| 2 Eye size and profile are small and flat (difficult to see). | 7 Tarsal terminal spurs are present (in both sexes). |
| 3 Punctations on scutum are not visible. | 8 Setae on scutum and alloscutum are numerous and long. |
| 4 Scutum posterior margin has a single concavity each side. | 9 Cervical fields are shallow and indistinct. |
| 5 Spiracle plate shape is circular. | |

FIGURE 65. *Rhipicephalus australis* (Australia cattle tick), female, dorsal and ventral.



- 1 Coxa 1 anterior spur is large and visible dorsally.
- 2 (Eyes of male are difficult to see by light microscopy).
- 3 Punctations on conscutum are not visible.
- 4 Lateral grooves are absent.
- 5 Setae on conscutum and alloscutum are long and numerous.

- 6 Fестоons are absent.
- 7 Columns of teeth on hypostome are 4 + 4.
- 8 Spiracle plate shape is circular (see figure for female).
- 9 Adanal plate shape is angular at posterior.
- 10 Caudal appendage is present in unfed male.
- 11 Anal groove is indistinct.

FIGURE 66. *Rhipicephalus australis* (Australia cattle tick), male, dorsal and ventral.

The males of these two species are distinguished as follows. Viewed dorsally, *R. australis* has an anterior projecting spur of coxa 1 that is visible; also *R. australis* has setae on the conscutum that are dense and long compared to the sparse and short setae of *R. sanguineus*. Ventrally, *R. australis* has adanal plates with an angular posterior margin, and 4 + 4 columns of teeth on the hypostome, whereas *R. sanguineus* has adanal plates with broadly curved posterior margins, and 3 + 3 columns of teeth on the hypostome. The anal groove of *R. australis* is indistinct compared to that of *R. sanguineus*. Male *R. australis* are 50% smaller than male *R. sanguineus*.

Hosts

Rhipicephalus australis is primarily a parasite of cattle in Australia, yet heavy infestations may also occur on horses, sheep, deer and water buffalo (Arundel & Sutherland 1988). Occasionally *R. australis* is found on marsupials, goats, cats, feral pigs and dogs (Roberts 1970). Larvae and young adults, especially males, may attach to humans but the local irritation and itching invariably leads to the tick being removed by the patient (Green 1971). But there is at least one case of a female attaching to and feeding to engorgement on a human and then producing viable eggs (Green 1971).

Life-cycle and seasonality

Rhipicephalus australis is a one-host tick, as are all five of the described species in the subgenus *Boophilus*. We presume the same applies to the two new species of *Boophilus*, which have not been described yet: *R. microplus*—Clade A species and *R. microplus*—Clade B species (Burger *et al.* 2014b). *Rhipicephalus australis* has a monotropic type of behaviour. The time spent by the three life-stages on the host is about three weeks and egg laying can be completed in about four weeks. In southeastern Queensland, large numbers of larvae are usually present on the vegetation in late spring, and successive generations of larvae then occur through the summer and into the cooler autumn and early winter months. There is a substantial literature on variation in seasonal abundance of ticks among regions of Queensland which is beyond the scope of this book. Interested readers might first read Seddon (1968) and Arundel and Sutherland (1988), and then consult the recent literature. Figure 6 is a drawing of the one-host life-cycle of *R. australis*.

Disease

Rhipicephalus australis transmits to cattle the protozoans *Babesia bovis* and *Ba. bigemina*, which cause bovine babesiosis, otherwise known as tick fever. *Babesia bovis* and *Ba. bigemina* infection is acquired during feeding by the adults of one generation of ticks. Then the *Babesia* infect the eggs of *R. australis in situ*, so larvae hatch with an infection of *Babesia* which they may pass to other cattle when the larvae feed on cattle. Thus, transmission of *Ba. bovis* and *Ba. bigemina* is trans-ovarial, which is also known as vertical transmission. *Rhipicephalus australis* also transmits to cattle the bacterium *Anaplasma marginale* which causes anaplasmosis in cattle, and *Borrelia theileri* which causes spirochaetosis in cattle. There is a large and growing literature on tick fever and anaplasmosis in Australia.

Heavy infestations of *R. australis* will directly lead to commercially important damage to hides by the formation of scar tissue (= granuloma) at the feeding sites. Experiments in Australia have shown that for each female tick that feeds to repletion there is a loss of 0.6 g to 0.9 g of potential growth by cattle (Sutherst *et al.* 1983, Jonsson 2006). Despite being nearly 50 years old, Seddon (1968 pp. 7–62) is still the most comprehensive and informative treatment of the ill effects of the feeding of *R. australis* on cattle in Australia.

Habitat and geographic distribution

Rhipicephalus australis is a tick of the tropics and sub-tropics. The precise geographic range of *R. australis* is not known but this tick definitely occurs in Australia, New Caledonia and Cambodia (Burger *et al.* 2014b). In Australia, *R. australis* occurs in a broad band following the general outline of the coast from northeastern New South Wales to northeastern Western Australia (Fig. 67). *Rhipicephalus australis* probably also occurs in the Philippines, Tahiti, Borneo, Sumatra, Java and Papua New Guinea (Estrada-Peña *et al.* 2012; Burger *et al.* 2014b), but this needs to be confirmed with *cox 1* nucleotide sequences. Doubtless, *R. australis* occurs elsewhere in Asia too.

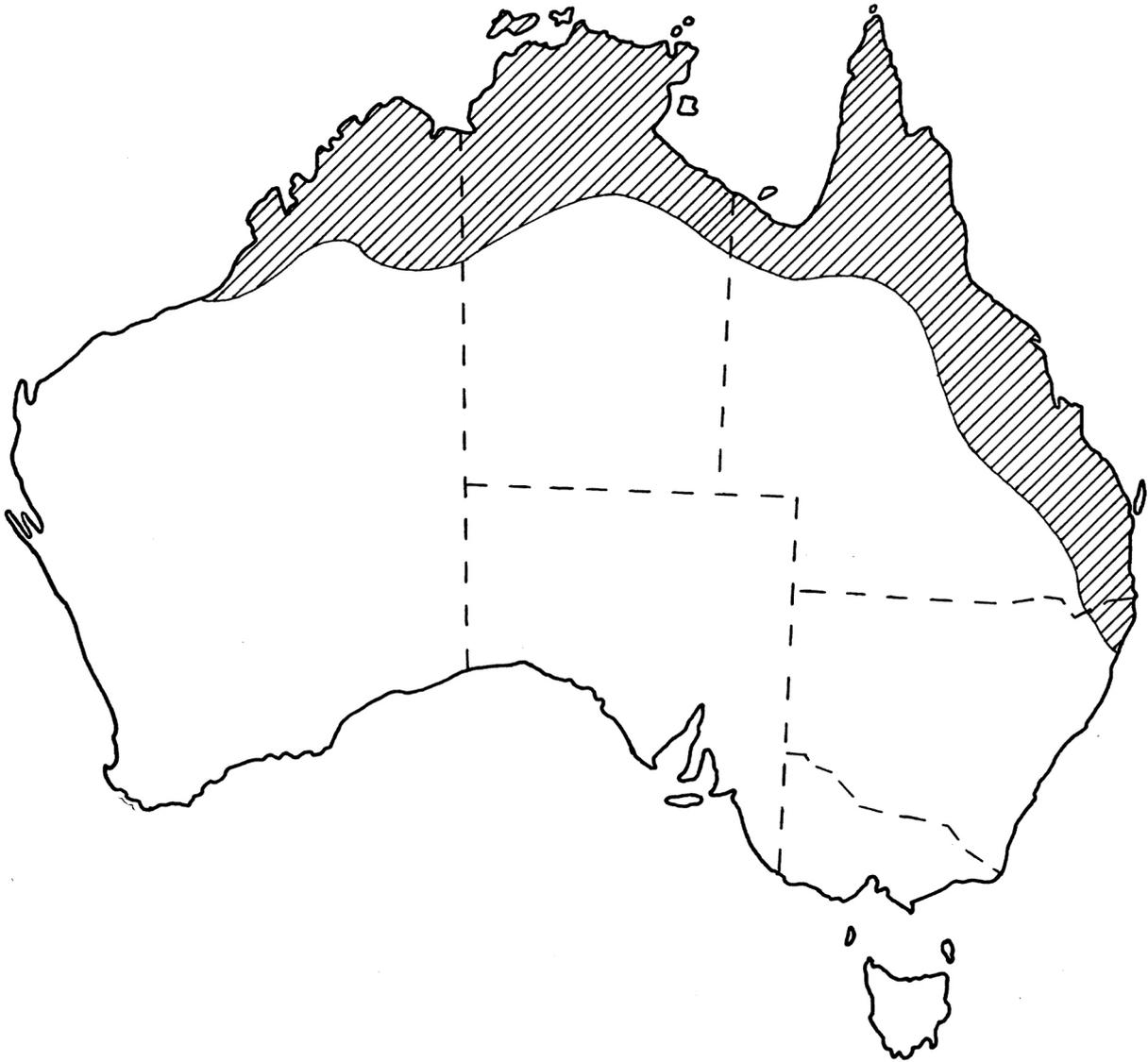


FIGURE 67. Geographic distribution of *Rhipicephalus australis* (Australia cattle tick) in Australia (redrawn from Arundel and Sutherland 1998).

Genes and genomes

The mitochondrial genome (Campbell & Barker, 1998, 1999, 2001; Burger *et al.* 2014b) and nuclear genes of *Rhipicephalus australis* have been studied more than those of most ticks. Before 2012 (Estrada-Peña *et al.* 2012), however, *R. australis* had the species name *microplus* (eg Crampton *et al.* 1998; Barker 1998; Chigagure *et al.* 2000; Murrell *et al.* 1999, 2000, 2001b) so care must be taken when using mitochondrial genomes and sequences in GenBank. *Rhipicephalus microplus* from Australia, where there has been much study, should be renamed in GenBank as *Rhipicephalus australis*.

Other information

There is a vast literature on the control, and attempted control, of *R. australis* in Australia. A review of this vast literature is beyond the scope of this book. Interested readers might first read Jonsson and Piper (2007), and then consult the recent literature. Murrell *et al.* (2003) cultured bacteria from *R. australis* and some other Australian ticks.

Taxonomy of the Australian cattle tick and history of its name. *Rhipicephalus australis* had the name *Boophilus microplus* until 2003 when the genus *Boophilus* was submerged into the genus *Rhipicephalus* (Murrell & Barker 2003). Thus, *Boophilus* became a subgenus of the genus *Rhipicephalus* which is indicated by parentheses

around the name *Boophilus*: *Rhipicephalus* (*Boophilus*). Then in 2012, the species-name "*australis*" was resurrected for cattle ticks in Australia, New Caledonia, and regions of south east Asia, including Philippines, Tahiti, Borneo, Sumatra, Java and Papua New Guinea (Estrada-Peña *et al.* 2012; see also Labruna *et al.* 2009). The name of the cattle tick in Australia thus reverted to the original name created in 1899 by Claude Fuller of what was then the Cape Colony, South Africa (Fuller 1899). Fuller was a South African expert on ticks who had been sent ticks from Australia for identification by C.J. Pound of Brisbane. Fuller gave the common name, the Queensland cattle tick, to the new species from Australia, *R. australis* Fuller, 1899. *Rhipicephalus australis* is one of at least four, but possibly more, species in the *Rhipicephalus* (*Boophilus*) *microplus* complex of species: *R. australis*, *R. annulatus*, *R. microplus*—Clade A species and *R. microplus*—Clade B species (Burger *et al.* 2014b). Our working hypothesis of the relationships of these four putative species (Fig. 68) has *R. australis* as the sister-species to *R. microplus*—Clade A species of Yunnan Province China, Brazil and Cambodia. Note that all four putative species in the *R. microplus* complex occur in Asia (Fig. 68). Detailed study of at least 798 bp, but ideally the entire (ca. 1500 bp), *cox1* DNA sequences is needed to resolve the geographic distribution of the ticks of the subgenus *Boophilus* in Asia (Burger *et al.* 2014b).

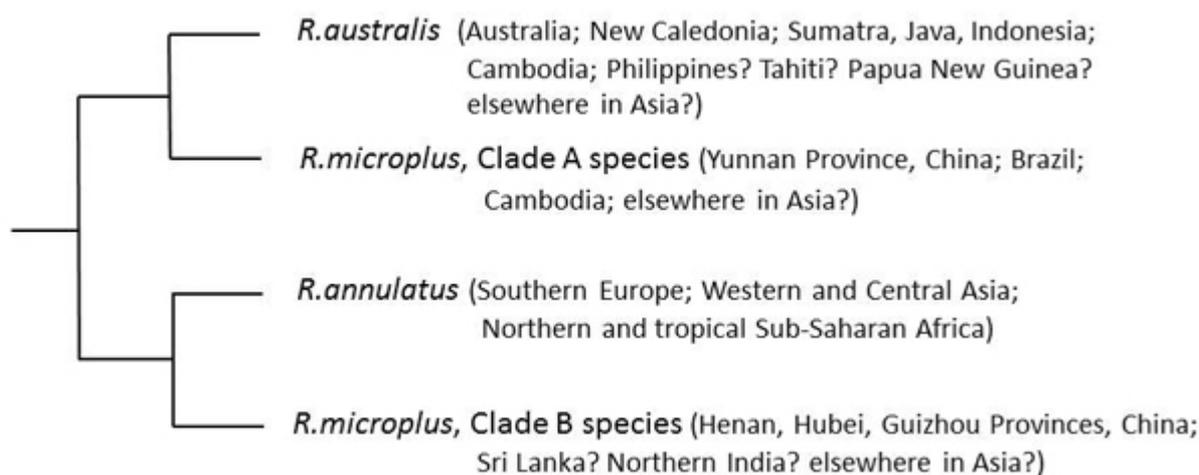


FIGURE 68. Proposed phylogenetic relationships of *Rhipicephalus australis* (Australian cattle tick), *R. annulatus* (cattle fever tick), *R. microplus*-Clade A species and *R. microplus*-Clade B species (after Burger *et al.* 2014b).

***Rhipicephalus sanguineus* (Latreille, 1806) (brown dog tick)**

General

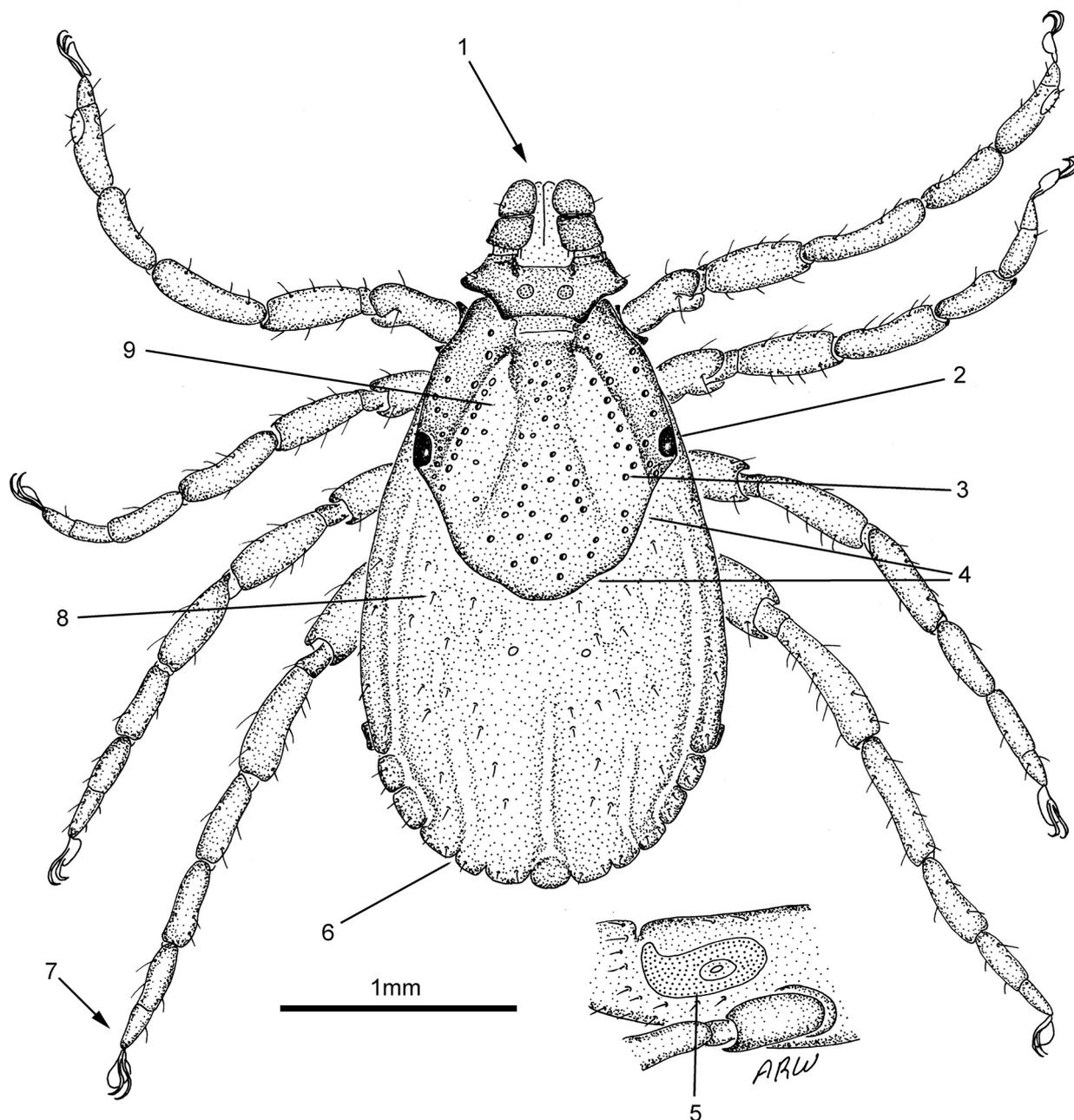
In Australia, *Rhipicephalus sanguineus* is known as the brown dog tick. Elsewhere, *R. sanguineus* is known as the kennel tick and the pan-tropical dog tick. *Rhipicephalus sanguineus* has become the most widespread tick in the tropics and sub-tropics because of its specialised feeding on domestic dogs.

Differential diagnosis

Rhipicephalus sanguineus is a small species of *Rhipicephalus*, usually of a dull yellow colour but some populations may be a mid brown colour. Elsewhere in the world, *R. sanguineus* has often been confused with other ticks in the *R. sanguineus* group of species. There are eight species in the *R. sanguineus* group of species: *R. bergeoni* Morel & Balis, 1976; *R. camicasi* Morel, Mouchet & Rodhain, 1976; *R. guilhoni* Morel & Vassiliades, 1963; *R. moucheti* Morel, 1965; *R. pusillus* Gil Collado, 1936; *R. sanguineus* (Latreille, 1806); *R. sulcatus* Neumann, 1908; *R. turanicus* Pomerantzev, 1940 (Pegram *et al.* 1987a) and probably some undescribed species as well (see for example Burlini *et al.* 2010, Moraes-Filho *et al.* 2011, Levin *et al.* 2012, Nava *et al.* 2012, Liu *et al.* 2013)

Rhipicephalus sanguineus can be differentiated easily from *R. australis*, the only other species of *Rhipicephalus* in Australia, using the following morphological features (Figs 69 & 70). Female *R. sanguineus* viewed dorsally, has a scutum that has: deep and distinct cervical fields, a double concavity on each side of the posterior scutum, and easily visible eyes. In contrast *R. australis* has a scutum with shallow and indistinct cervical

fields, a single concavity on each side of the posterior scutum, and eyes that are difficult to see. The setae on the scutum and alloscutum of *R. sanguineus* are sparse and short whilst those of *R. australis* are dense and long. On lateral and ventral surfaces of females, *R. sanguineus* has large comma-shaped spiracle plates and a hypostome with 3 + 3 columns of teeth. In contrast, *R. australis* has small circular spiracle plates and 4 + 4 columns of teeth on the hypostome.



1 Columns of teeth on hypostome are 3 + 3 (see figure for male).

2 Eye size and profile are large and slightly convex.

3 Punctations on scutum are conspicuous.

4 Scutum posterior margin has a double concavity each side.

5 Spiracle plate shape is like a comma.

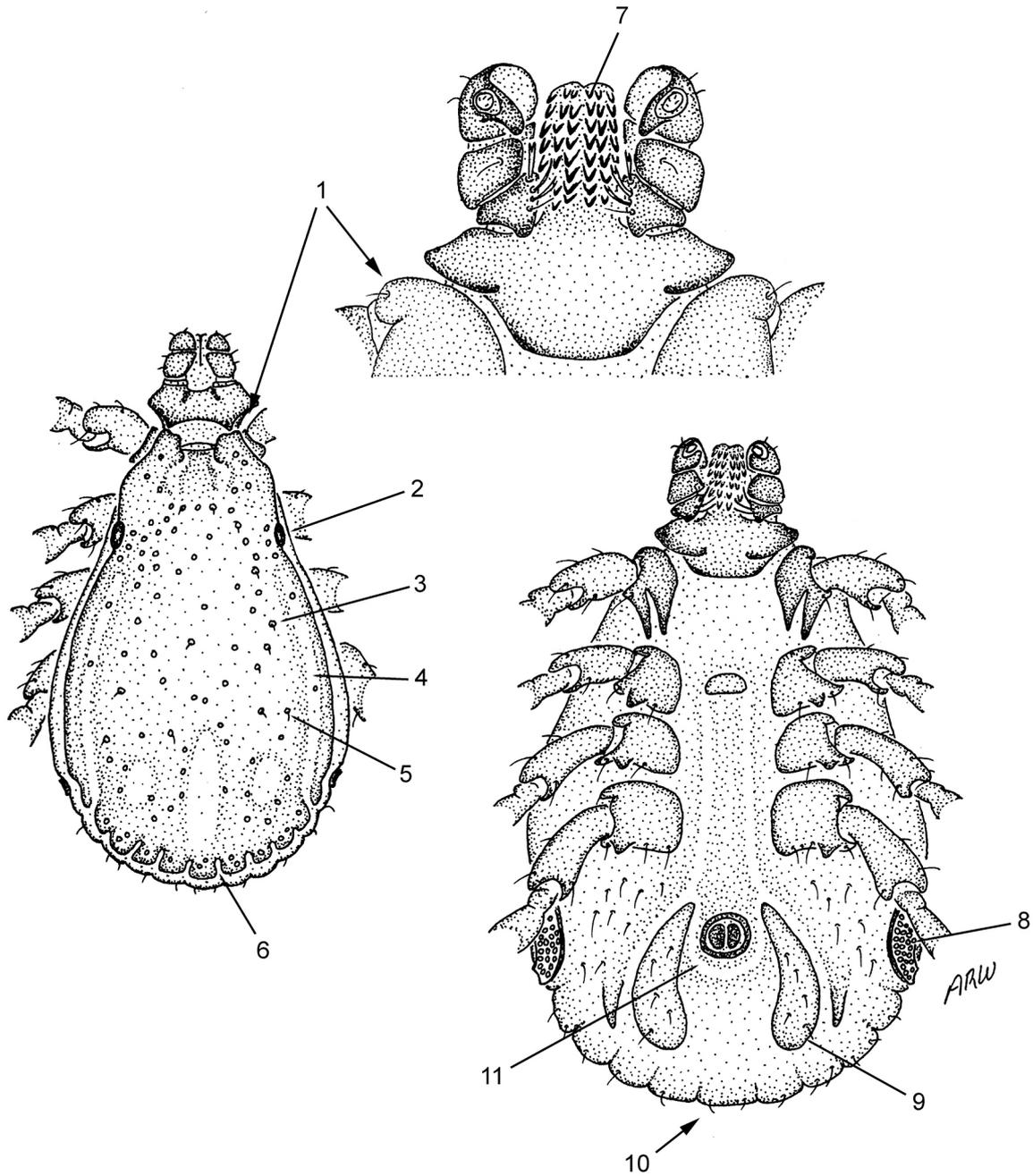
6 Festoons are present.

7 Tarsal terminal spurs are absent (in both sexes).

8 Setae on scutum and alloscutum are short and sparse.

9 Cervical fields are deep and distinct.

FIGURE 69. *Rhipicephalus sanguineus* (brown dog tick), female, dorsal.



- 1 Coxa 1 anterior spur is small and not visible dorsally.
- 2 Eye size and profile is large and slightly convex.
- 3 Punctations on conscutum are conspicuous.
- 4 Lateral grooves are present.
- 5 Setae on conscutum and alloscutum are short and sparse.

- 6 Festoons are present.
- 7 Columns of teeth on hypostome are 3 + 3.
- 8 Spiracle plate shape is like a comma (see figure for female).
- 9 Adanal plate shape is broadly curved at posterior.
- 10 Caudal appendage is absent in unfed male.
- 11 Anal groove is distinct.

FIGURE 70. *Rhipicephalus sanguineus* (brown dog tick), male, dorsal and ventral.

There are similar differences between the males. Viewed dorsally, *R. sanguineus* does not have any anterior projecting spur on coxa 1; also it has setae on the conscutum that are sparse and short whereas *R. australis* has setae that are dense and long. Ventrally, *R. sanguineus* shows adanal plates that are broadly curved at their posterior margin and a hypostome with 3 + 3 columns of teeth, whereas *R. australis* shows adanal plates with an angular posterior margin and a hypostome with 4 + 4 columns of teeth. The anal groove of *R. sanguineus* is distinct compared to that of *R. australis*. Male *R. sanguineus* are 50% larger than male *R. australis*.

Hosts

Domestic dogs (*Canis familiaris*) are the host for which *R. sanguineus* is specialised, but this tick may be found on cattle too. Dogs are hosts for all of the life stages. The behaviour of this tick is both domestic and monotropic: larvae, nymphs and adults feed on dogs. Adults attach on the ears, neck and shoulders, nymphs are also found on the ears and shoulders, and larvae attach particularly to the belly and flanks of dogs. Immature stages of this tick attempt to attach to humans. Hosts other than dogs are usually only infested when dogs are not present to maintain tick populations of this tick. Curiously, there are no published records of *R. sanguineus* on *Canis lupus*, the dingo. Roberts (1965) was the first to be struck by this observation. *Rhipicephalus sanguineus* has, however, been found on “camp dogs” in Australian aboriginal communities (Barker & Campelo, unpublished data).

Life-cycle and seasonality

Rhipicephalus sanguineus is a three-host tick as are most species of the genus *Rhipicephalus* (with the exceptions of *R. bursa*, *R. evertsi*, *R. glabroscutatum*, and the species of the subgenus *Boophilus*) (Murrell *et al.* 2001a). The engorged female detaches and lays approximately 3,200 eggs within seven to 28 days of detaching from a dog. The eggs hatch within three to 10 weeks. The larvae engorge in three to eight days and moult within two to six weeks. The nymphs engorge in four to 10 days and moult within two to 26 weeks. The females may engorge in seven days but they can stay on the dog for three weeks if unmated and the males remain on the dog for several months for repeated matings. The life-cycle can be completed in 10 weeks under ideal conditions. More than one life-cycle per year is possible. Although this tick can survive in open environments, it is highly adapted to living in dog kennels and in the homes of humans. Thus, *R. sanguineus* is atypical of *Rhipicephalus* ticks which are usually exophilic (live outdoors). The females climb up walls and lay eggs in cracks and crevices in walls or they may lay eggs under the dog's bedding or in nearby cracks and crevices. The larvae and nymphs usually moult in the same sites as the females lay their eggs. Dogs that are tied up repeatedly in the same kennel may become heavily infested. In artificially heated homes the tick's feeding activity may extend into winter.

Disease

Rhipicephalus sanguineus may harbour the bacterium *Ehrlichia canis* which may cause canine ehrlichiosis which is also known as canine tropical pancytopenia (Groves *et al.* 1975). Infection with *E. canis* may develop into a chronic form of the disease. *Rhipicephalus sanguineus* may harbour the bacterium *Rickettsia conorii* which may cause boutonneuse fever (= Mediterranean spotted fever) and Israeli spotted fever (Levin *et al.* 2012); and *Ri. rickettsii*, which may cause Brazilian spotted fever (Ogrzewalska *et al.* 2012). *Rhipicephalus sanguineus* may transmit the protozoans *Babesia vogeli* and *B. gibsoni* to dogs and this may cause canine babesiosis (Shortt 1973). The protozoan *Hepatozoon canis* may be transmitted from ticks to dogs when dogs swallow ticks during grooming; this pathogen may cause hepatozoonosis in dogs.

Habitat and geographic distribution

Rhipicephalus sanguineus is common between about latitudes 50° N and 30° S throughout the world. In Australia, too, *R. sanguineus* is most common north of latitude 30° S although it is occasionally found as far as south as Sydney and Melbourne (Roberts 1952, 1965). *Rhipicephalus sanguineus* may be more common on dogs from Aboriginal communities and townships than in other towns and cities in Australia (Barker unpublished anecdotal observations); this idea needs to be tested. More data on the southern boundary of the geographic distribution postulated by Roberts (1965a, Fig. 5) are also needed.

Genes and genomes

The genes of *Rhipicephalus sanguineus* have been studied more than those of most ticks; 1377 sequences were in

GenBank and thus available to the public at the time of writing, including the entire mitochondrial genome (14, 710 bp; Black & Roehrdanz 1998).

Other information

See Burger *et al.* (2013) and references for hypotheses for the evolutionary history (phylogeny) of *R. sanguineus*.

Summation and future prospects

Hoogstraal (1971) lamented the "slight amount of available biological information on Australian ticks" in the book *Australian Ticks* (Roberts 1970). Much has changed since "Bob" Roberts drafted his book in the late 1960s. Indeed, the next decade, the 1970s, was in our view a golden decade of research on the hosts, life-cycles, seasonality, and diseases associated with tick infestation. The golden city of this decade was Brisbane. Substantial and well-funded groups in two divisions of the CSIRO (the Divisions of Tropical Animal Production, and the Division of Entomology), the Queensland Department of Primary Industries, and the Department of Parasitology in the Faculty of Science at The University of Queensland published many seminal papers on ticks in the 1970s. Almost all of these papers were on *Ixodes holocyclus* (paralysis tick), *Haemaphysalis longicornis* (bush tick) and *Rhipicephalus australis* (Australian cattle tick). All three of these ticks flourish in certain moist and sub-tropical environments near Brisbane. So Brisbane was the logical and perhaps only feasible place to unmask, so completely, the biology of these three species of ticks. Of course, the paralysis tick, Australian cattle tick and to a much less extent, the bush tick, are the three villains of the Australian tick fauna: the Australian cattle tick is a major limiter of cattle production in northern Australia whereas the paralysis tick kills many dogs each year and is *the* main tick that attaches to people along the east coast of Australia. Indeed, in the experience of one of us (SB) most ticks found on people along the eastern coast of Australia are paralysis ticks. To give our readers some idea of the scope of the research on these three tick species in Brisbane in the 1970s we list, in alphabetical order, the authors of papers on these ticks that were published between 1970 and 1979, drawn from our list of references: P.E Bird, Keith C.K. Binnington, Bernard M. Doube, Alan C.G. Heath, Peter E. Green, Dave H. Kemp, Douglas E. Moorhouse, J.A. Roberts, Bob W. Sutherst, Klaus B.W. Utech, R. Harry Wharton and B.M. Wagland. Times change. Indeed the site of the Long Pocket Laboratories and tick-pens of the CSIRO Division of Tropical Animal Production is soon to be a residential suburb. But tick research continues in Brisbane, particularly, in the Queensland Department of Primary Industries (now the Department of Employment, Economic Development and Innovation) and in the Department of Parasitology of the Faculty of Science at The University of Queensland.

So what about the future? Our reading of 500 or so papers for this book has revealed many gaps in our knowledge and conceptual understanding of Australian ticks. First and foremost we need hundreds, probably thousands, of new site-records and host-records of ticks so that the climatic, vegetation, soil and host factors that determine which tick lives here and not there can be discovered. At present we don't even have enough published site-localities to draw an accurate map of the geographic distribution of many of the 16 species of ticks in this book; let alone the 54 other species of Australian ticks. Contrast the situation in Australia with the exhaustive knowledge of *Ixodes ricinus* in Europe and *I. scapularis* in North America. The work of Laan, Handasyde & Beveridge on *Haemaphysalis bancrofti* (wallaby tick) and *Ixodes hirsti* (Hirst's marsupial tick) are fine exemplars (Laan *et al* 2011a, 2011b).

Second, many taxonomic questions remain as they always will if we continue to study nature. For example, *I. tasmani* may well be a complex of species rather than a single species (Roberts 1970) and the poultry-infesting *Argas* of the New World may be a species complex too (see 'Other information' in the species account of *A. robersti*). These are many fine PhD research topics on offer in the taxonomy of Australian ticks.

Third, why do closely related ticks differ in their morphology? For example, of the four *Ixodes* species in this book, the adult female *I. tasmani* has a conspicuously different type of mouthparts to the other three species of *Ixodes*. The sensory palps of *I. tasmani* have unusually wide and splayed first and third articles, and the piercing and blood-sucking structures, comprising cheliceral sheaths and hypostome, are unusually short and stubby. What type of adaptation to the range of hosts of *I. tasmani*, and other *Ixodes* species, might this represent?

Fourth, there is tremendous scope for studies of the hosts, life-cycles and seasonality, and of the diseases associated with the 54 species of ticks that infest only wild mammals, birds and reptiles in Australia. Many of these

ticks are amenable to laboratory study, or at least to study in outdoor wildlife enclosures (eg Gemmell et al 1991), since these ticks live on locally abundant wildlife that are often found dead on the side of the road and/or are easily captured. Take for example, *Ixodes trichosuri*, a tick of the common brushtail possum, *Trichosurus vulpecula*, which is the most common native animal in suburban Australia. Brushtail possums readily enter man-made nest-boxes and thus their ticks might be studied and even raised in nest-boxes and the laboratory, without too much trouble, if the larvae, nymphs and adults are allowed to feed on their natural host, the brushtail possum. The work of Murdoch & Spratt (2005) on *I. tasmani* (common marsupial tick) is a fine exemplar.

Lastly, what are the main drivers of speciation in Australian ticks: changes in the climate of Australia and/or changes in the fauna (hosts of ticks)? Elsewhere, we consider the extraordinary changes in the global position and thus latitude and climate of that part of the supercontinent, Gondwana, that became Australia, since ticks first evolved 300 to 400 million years ago in the Carboniferous–Devonian (Barker, Walker & Campelo 2014). These changes in latitude and climate were associated with extraordinary changes in the fauna of Australia, that is, the *hosts* of the ancestors of the 65 species of endemic Australian ticks. Indeed, almost all of the original hosts of these ticks are now extinct! Australian ticks, like other ticks, are extraordinarily adaptable and thus were able to survive the extinction of their hosts.

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