

Volume 33

August 2014

# CONSULTING ECOLOGY



[www.ecansw.org.au](http://www.ecansw.org.au)

ISSN 1836-6813

Newsletter of the Ecological Consultants Association of NSW

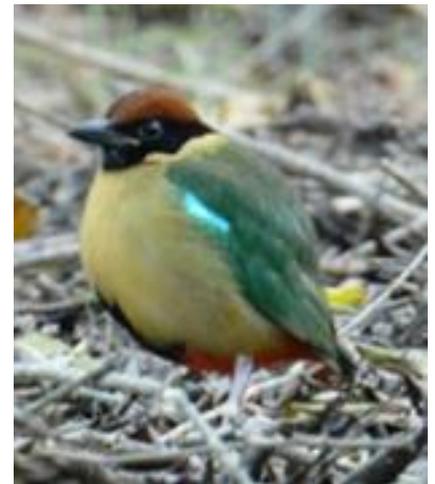


**Southern Myotis in a nest box** (Photo Courtesy of Veronica Silver) - see page 18.

**VOLUME 33 August 2014**

**INSIDE THIS ISSUE!**

- 1 ECA Office Bearers 2014-2015
  - 1 Message from the President
  - 2 Euroky: Recent microbat taxonomic revision and name changes - the *Mormopterus* genus
  - 5 Upcoming Events in 2014
  - 5 August 2014 ECA Membership Report
  - 6 Recent Literature and New Publications
  - 6 2014 Ecological Consultants Association of NSW Annual Conference
  - 7 Photo Competition
  - 9 Getting more from your bat surveys
  - 15 Creating Roosting Habitat Alternatives for Breeding Myotis
  - 18 Holidaying or Moving in?: The story of the most southerly confirmed record of Noisy Pitta (*Pitta versicolor*) at Morunna Point, Wallaga Lake, NSW far south coast.
  - 21 Traps, Transects and Technology: Detectability of the threatened Rosenberg's Goanna (*Varanus rosenbergi*)
  - 25 ECA Photo Gallery: Photo Competition Entries
  - 27 Field Work Ticks Me Off
  - 50 The platypus and the environmental impact assessment process: more cogitations of a consultant.
  - 64 Advertising Opportunities with the ECA
  - 64 Contributions to the Newsletter, Volume 34
- Back cover ECA Photo Gallery: Photo Competition Entries



**Noisy Pitta observed at Morunna Point, Wallaga Lake, NSW far south coast** (Photo courtesy of Steve Sass, EnviroKey). See page 19



**Rosenberg's Goanna** (Photo courtesy and copyright Stuart Cooney). See Page 22

**Editor: Jason Berrigan**

**Design and Layout: Amy Rowles**

**Front Cover Photo: Waratah on Newnes Plateau, Lithgow. Photo Courtesy and Copyright of Phil Cameron.**

## ECA Office Bearers 2014-2015

**President:**

Martin Denny  
[president@ecansw.org.au](mailto:president@ecansw.org.au)

**1<sup>st</sup> Vice-President:**

Belinda Pellow  
[belinda.pellow@austmus.gov.au](mailto:belinda.pellow@austmus.gov.au)

**2<sup>nd</sup> Vice-President:**

Alison Hunt  
[alison@ahecology.com](mailto:alison@ahecology.com)

**Secretary:**

Adam Greenhalgh  
[secretary@ecansw.org.au](mailto:secretary@ecansw.org.au)

**Treasurer:**

Andrew Lothian  
[treasurer@ecansw.org.au](mailto:treasurer@ecansw.org.au)

**Councillors:**

Paul Burcher  
[paul@aquilaeco.com.au](mailto:paul@aquilaeco.com.au)  
Rhidian Harrington  
[rharrington@niche-eh.com](mailto:rharrington@niche-eh.com)  
Deryk Engel  
[deryke@lesryk.com.au](mailto:deryke@lesryk.com.au)  
Toby Lambert  
[LambertT@pbworld.com](mailto:LambertT@pbworld.com)  
Matt Richardson  
[mrichardson@niche-eh.com](mailto:mrichardson@niche-eh.com)  
John Travers  
[info@traverseecology.com.au](mailto:info@traverseecology.com.au)  
Danny Wotherspoon  
[wilderness@mountains.net.au](mailto:wilderness@mountains.net.au)  
Isaac Mamott  
[isaac.mamott@sclerophyll.com.au](mailto:isaac.mamott@sclerophyll.com.au)  
Jane Murray  
[jmurray@biosis.com.au](mailto:jmurray@biosis.com.au)  
Kristy Peters  
[kpeters@kleinfelder.com](mailto:kpeters@kleinfelder.com)

**Administration Assistant:****Membership Officer:**

Amy Rowles  
[admin@ecansw.org.au](mailto:admin@ecansw.org.au)  
39 Platt St, Waratah, NSW, 2298

**Newsletter Editor:**

Jason Berrigan  
[jason.berrigan@naturecall.com.au](mailto:jason.berrigan@naturecall.com.au)

## Message from the President

Dear members,

Welcome to a new year of the ECA (not the annual, financial or Chinese new year but the beginning of the new Council). With the increasing membership and established profile, the ECA makes an important contribution to the process of environmental impact and conservation. It is hoped that much will be achieved during 2014-2015. After another successful and informative conference it is time to plan for the future and a series of workshops, as well as the annual conference are already in the planning stages. The Council is determined to proceed with a Certification scheme for ecological consultants - this is long overdue and is being demanded by politicians as well as government and non-government agencies. The support of all members is essential if Certification is to be achieved.

I notice the Australian Department of Environment has just released a "Species of National Environmental Significance Database" which contains map summaries which provide general information on the distribution of plant and animal species related to the EPBC Act. Species covered by the database include threatened and migratory species. If we start to add to this database with that obtainable from OEH, Botanical Gardens, VIS, Atlas of Living Australia etc etc we have a vast array of distributional information about flora and fauna that allows an ecological consultant to develop a species list for an area without leaving the computer. Is this the future? The blossoming of electronic gadgetry has already provided a wealth of information and techniques to the ecologist's arsenal. On my iPhone I have an app that provides all the 1:25,000 topographic maps for NSW so that I can locate myself and the survey sites at any time. I also have apps for recording grid references, measuring the height of trees and providing distributional information and calls of Australian birds. There are now hundreds of such additions to a phone or iPod that can provide information and methodology unheard-of in past decades - in a box the size of a deck of cards we carry 100s of years of knowledge.

If we add to this range of equipment the use of remote cameras, bat ultrasonic call detectors, thermal imaging, satellite image interpretation, drones, audio recorders - the list goes on - it is coming to a situation that it may not be necessary for an ecologist to enter the field. All will be available from databases, modelled distributions and habitat preferences and information coming from remotely placed recorders. Who knows? We may be known in the future as the digital ecological consultants!

In the mean time let us still enjoy the hands on role we are able to take and have a good year.

*Martin Denny*

# EUROKY

**Euroky: ability of an organism to adapt to changes in the environment**

*If you have any interesting observations or useful hints and information that you would like to share in the euroky column, please forward them to the newsletter editor or administration assistant to be included in the next edition.*

## RECENT MICROBAT TAXONOMIC REVISION AND NAME CHANGES - THE MORMOPTERUS GENUS

Anna McConville

The long-awaited taxonomic revision of the *Mormopterus* genus of microbats in Australia is here! The results of Terry Reardon's study, which combines morphological and genetic analyses, have just been published in the Australian Journal of Zoology (Reardon, McKenzie *et al* 2014). So I thought I'd give you a quick summary and a guide to the major name changes that have arisen from this much needed research.

### A bit of background

As you may be aware, the taxonomy of the *Mormopterus* genus in Australia has been an ongoing source of confusion. Reviews of the taxonomic history of Australian *Mormopterus* in the late 1980's tentatively concluded that there were three *Mormopterus* species in Australia: *M. norfolkensis*, *M. planiceps* and *M. beccarii* (Allison 1989; Mahoney 1988). This was soon followed by a genetic study on Australian Molossidae, which determined that at least six *Mormopterus* species occurred in Australia (Adams, Reardon *et al* 1988), referred to as *M. species 1* through to *M. species 6*. *Mormopterus norfolkensis* was not included in the study by Adams, Reardon *et al* (1988), so essentially from the start of the 1990's, it was recognised that at least seven Australian *Mormopterus* species: *Mormopterus*

*norfolkensis* and *M. species 1* through to *M. species 6*.

In the 25 years since these early studies, many different informal names have been given to Australian *Mormopterus* species in field guides and other published material. This has created a great deal of confusion among Australian ecological professionals and has made the preparation of species lists and field identification frustrating.

### So what do we call them now?

Australian *Mormopterus* have been found to be different enough from overseas *Mormopterus* to warrant three different subgenera (Reardon, McKenzie *et al* 2014).

**Family:** Molossidae

**Genus:** *Mormopterus*

**Subgenera:** *Micronomus*, *Setirostris* and *Ozimops*

A total of nine *Mormopterus* species are now recognised to occur in Australia. The new scientific and common names and how these relate to old names used in key literature and field guides are outlined in Table 1. If you are still confused, the distribution maps for each species and further description can be found in the journal article (Reardon, McKenzie *et al* 2014).

You don't need to report the subgenus name (in brackets) if you prefer to keep things simple; but with fantastic names like *Ozimops*, I think I will be using them. Also, the same abbreviation rules apply for subgenera as for genera, in that you can represent the subgenus name by the first letter in subsequent uses. As an example using my favourite species, *Mormopterus (Micronomus) norfolkensis*, would be shortened to *M.(M.) norfolkensis*.

These new names will probably take a little while to trickle through to things like the Atlas of NSW Wildlife and the legislation, but I recommend that you start using these new names (scientific and common) in your upcoming reports and record submissions to avoid continued confusion.

Table 1 : Current names of former Australian *Mormopterus* species, contrasted with names used in other literature (adapted from Reardon, McKenzie *et al.* 2014)

Current Scientific Name <sup>†</sup>	Current Common Name <sup>†</sup>	Adams, Reardon <i>et al.</i> (1988)	Reardon, Adams <i>et al.</i> (2008)	Van Dyck and Strahan (2008)	Churchill (2008)	Hall (2009)
<i>Mormopterus (Micronomus) norfolkensis</i>	East coast free-tailed bat	N/A	<i>Mormopterus norfolkensis</i>	<i>Mormopterus norfolkensis</i>	<i>Micronomus norfolkensis</i>	<i>Mormopterus norfolkensis</i>
<i>Mormopterus (Setirostris) eleryi</i>	Bristle-faced free-tailed bat	<i>Mormopterus</i> Species 6	<i>Mormopterus eleryi</i>	<i>Mormopterus</i> sp.	<i>Mormopterus</i> species 6	
<i>Mormopterus (Ozimops) cobourgianus</i>	North-western free-tailed bat	<i>Mormopterus</i> Species 5 Populations U & V	N/A	<i>Mormopterus loriae cobourgiana</i>	<i>Mormopterus corbourgiana</i>	<i>Mormopterus</i> sp.
<i>Mormopterus (Ozimops) halli</i>	Cape York free-tailed bat	<i>Mormopterus</i> Species 5 Populations S & T	N/A	<i>Mormopterus loriae ridei</i>	<i>Mormopterus ridei</i>	<i>Mormopterus</i> sp.
<i>Mormopterus (Ozimops) kitcheneri</i>	South-western free-tailed bat	<i>Mormopterus</i> Species 4 Population O	N/A	<i>Mormopterus</i> sp.	<i>Mormopterus</i> species 4	<i>Mormopterus planiceps</i>
<i>Mormopterus (Ozimops) lumsdenae</i>	Northern free-tailed bat	<i>Mormopterus</i> Species 1	N/A	<i>Mormopterus beccarii astrolabiensis</i>	<i>Mormopterus beccarii</i>	<i>Mormopterus beccarii</i>
<i>Mormopterus (Ozimops) petersi</i>	Inland free-tailed bat	<i>Mormopterus</i> Species 3	N/A	<i>Mormopterus</i> sp.	<i>Mormopterus</i> species 3	<i>Mormopterus planiceps</i>
<i>Mormopterus (Ozimops) planiceps</i>	South-eastern free-tailed bat	<i>Mormopterus</i> Species 4 Populations P, Q & R	N/A	<i>Mormopterus</i> sp.	<i>Mormopterus</i> species 4	<i>Mormopterus planiceps</i>
<i>Mormopterus (Ozimops) ridei</i>	Eastern free-tailed bat	<i>Mormopterus</i> Species 2	N/A	<i>Mormopterus</i> sp.	<i>Mormopterus ridei</i>	<i>Mormopterus planiceps</i>

<sup>†</sup> Based on Reardon, McKenzie *et al.* (2014)

# TITLEY SCIENTIFIC

Science, Technology and Innovation

- ♦ Acoustic detection ♦
- ♦ Radio transmitters ♦
- ♦ Telemetry receivers ♦
  - ♦ Antennas ♦
  - ♦ Camera traps ♦
  - ♦ Trap alerts ♦



## ANABAT EXPRESS



- Weatherproof
- Omnidirectional microphone
- In-built GPS
- 14 nights on 4 AAs
- Records temperature
- Automatic night-only mode

**\$850** (exc. GST)

[www.titley-scientific.com](http://www.titley-scientific.com)

Or call (07) 3205 8450

Free shipping  
with codeword:  
**ECA**

Also, if you aren't aware of the amazing diversity in bat genitalia, check out the manuscript for some amazing photographs and to find out how they are used to identify the different *Mormopterus* species.

### References

Adams, M., Reardon, T., Baverstock, P., and Watts, C. (1988) Electrophoretic resolution of species boundaries in Australian Microchiroptera. IV. The Molossidae (Chiroptera:Vespertilionidae). *Australian Journal of Biological Science* **41**, 315-26.

Allison, F.R. (1989) Molossidae. In 'Fauna of Australia. 1B. Mammalia.' (Eds. DW Walton and BJ Richardson) pp. 21-88. (Australian Government Publishing Service: Canberra)

Churchill, S. (2008) 'Australian Bats.' Second edn. (Allen & Unwin: Crows Nest)

Hall, L. (2009) 'A Wild Australia Guide: Bats.' (Steve Parish Publishing: Queensland)

Mahoney, J.A., Walton, D.W. (1988) Molossidae. In 'Zoological Catalogue of Australia.' (Ed. DW Walton) pp. 146-150. (Australian Government Publishing Service: Canberra)

Reardon, T., Adams, M., McKenzie, N., and Jenkins, P. (2008) A new species of Australian freetail bat *Mormopterus eleryi* sp. nov. (Chiroptera: Molossidae) and a taxonomic reappraisal of *M. norfolkensis* (Gray). *Zootaxa* **1875**, 1-31.

Reardon, T.B., McKenzie, N.L., Cooper, S.J.B., Appleton, B., Carthew, S., and Adams, M. (2014) A molecular and morphological investigation of species boundaries and phylogenetic relationships in Australian free-tailed bats *Mormopterus* (Chiroptera : Molossidae). *Australian Journal of Zoology* **62**(2), 109-136.

Van Dyck, S., and Strahan, R. (2008) 'The Mammals of Australia.' Third edn. (Reed New Holland: Sydney)

## Upcoming Events in 2014

### ECA Events

#### • PROPOSED ECA WORKSHOPS

2014 - 2015

- ◆ **Business Development and Practices Workshop**
- ◆ **Fungi : Ecology, Identification and conservation status**
- ◆ **Shorebirds**
- ◆ **Rainforest Plant ID**

The dates and venues for these workshops are yet to be determined. You may register your interest in any of these workshops by emailing [admin@ecansw.org.au](mailto:admin@ecansw.org.au).

#### • **Climate Adaptation 2014 : Future Challenges**

*Date:* 30th September - 2nd October 2014

*Location:* Gold Coast

*Details:* <http://www.nccarf.edu.au/conference2014/>

#### • **APCC10: 10th Australasian Plant Conservation Conference 2014 (The Australian Network for Plant Conservation Inc. )**

*Date:* 11-14th November 2014

*Location:* Hobart

*Details:* <http://www.anbg.gov.au/anpc/conferences/2014/>

#### • **Territory Natural Resource Management 2014 Conference**

*Date:* 18th-20th November 2014

*Location:* Darwin

*Details:* <http://www.territorynrm.org.au/2014-conference/>

#### • **IUCN World Parks Congress**

*Date:* 12-19th November 2014

*Location:* Sydney

*Details:* <http://worldparkscongress.org/index.html>

#### • **Australian Rangelands Society 18th Biennial Conference: Innovation in the Rangelands**

*Date:* 12-16th April 2015

*Location:* Alice Springs

*Details:* <http://www.austrangesoc.com.au/site/>

#### • **2014 RZS NSW Forum: The value of protected areas for fauna conservation**

*Date:* 8th November 2014

*Location:* Australian Museum

*Details:* <http://rzsnsw.org.au/>

### August 2014 ECA Membership Report

*Amy Rowles*

*ECA administrative assistant*

In total we have 161 members, comprised of 127 Practising Ecological Consultants, 3 Associate (Consultants), 16 Associate (Government Ecological/Environment Officer), 12 Associate (Non-practising), 2 Associate (Subscriber) and 1 Student. We have had fifteen new members and five current applicants over the last six months. The new members are introduced below:

- Rowena Hamer
- Anna McConville
- David Bain
- David Russell
- Mia Dalby-Ball
- De-Anne Attard
- Jacob Sife
- Anthea Whitlam
- Carl Corden
- Rebecca Southwell
- Timothy Johnson
- Rebecca Burley
- Alexander Wray-Barnes
- Daniel Whaite

Members may email any ideas for future ECA workshop topics or conference themes to Amy Rowles [admin@ecansw.org.au](mailto:admin@ecansw.org.au)

## Recent Literature and New Publications

### Recent Journal Articles / Literature

Hewitt A. et al (2014) **Plant-level fecundity and andromonoecy in three common (*Melaleuca styphelioides*, *M. thymifolia*, *M. nodosa*) and one rare (*M. deanei*) *Melaleuca* (Myrtaceae) species of the Sydney region.** *Australian Journal of Botany* **62(4)**:276-285 - <http://dx.doi.org/10.1071/ZO13090>

You Li M. et al (2014) **Delineation of conservation units in an endangered marsupial, the southern brown bandicoot (*Isodon obesulus obesulus*), in South Australia / western Victoria, Australia.** *Australian Journal of Zoology* - <http://dx.doi.org/10.1071/ZO14038>

Serena M. et al (2014) **Variation in platypus (*Ornithorhynchus anatinus*) life-history attributes and population trajectories in urban streams.** *Australian Journal of Zoology* **62(3)**: 223-234 <http://dx.doi.org/10.1071/ZO13079>

Taylor B. and Goldingay R. (2014) **Use of highway underpasses by bandicoots over a 7-year period that encompassed road widening.** *Australian Mammalogy* **36(2)** 178-183 <http://dx.doi.org/10.1071/AM13034>

Mezler A. et al. (2014) **The habitat and diet of koalas (*Phascolarctos cinereus*) in Queensland.** *Australian Mammalogy* **36(2)**: 189-199 <http://dx.doi.org/10.1071/AM13032>

Harris J., Goldingay R. and O'Brooks L. (2014) **Population ecology of the eastern pygmy-possum (*Cercartetus nanus*) in a montane woodland in southern New South Wales.** *Australian Mammalogy* **36(2)** 212-218 <http://dx.doi.org/10.1071/AM13044>

Goldingay R. (2014) **Gliding performance in the yellow-bellied glider in low-canopy forest.** *Australian Mammalogy* **36(2)** 254-258 <http://dx.doi.org/10.1071/AM14006>

Davis A., Major R. and Taylor C. (2014) **Distribution of tree-hollows and hollow preferences by parrots in an urban landscape.** *Emu* - <http://dx.doi.org/10.1071/MU13065>

Old J., Spencer R. and Wolfenden J. (2014) **The Common Myna (*Sturnus tristis*) in urban, rural and semi-rural areas in Greater Sydney and its surrounds.** *Emu* **114(3)** 241-248 <http://dx.doi.org/10.1071/MU13029>

Nugent D., Leonard S. and Clarke M. (2014) **Interactions between the superb lyrebird (*Menura novaehollandiae*) and fire in south-eastern Australia** *Wildlife Research* - <http://dx.doi.org/10.1071/WR14052>

Kirkwood R. et al (2014) **Lessons from long-term predator control: a case study with the red fox .** *Wildlife Research* - <http://dx.doi.org/10.1071/WR13196>

Quy R. et al. (2014) **Effects of a GnRH vaccine on the movement and activity of free-living wild boar (*Sus scrofa*).** *Wildlife Research* - <http://dx.doi.org/10.1071/WR14035>

Bengsen A. (2014) **Effects of coordinated poison-baiting programs on survival and abundance in two red fox populations.** *Wildlife Research* - <http://dx.doi.org/10.1071/WR13202>

Olsen J. et al (2014) **Is wedge-tailed eagle, *Aquila audax*, survival and breeding success closely linked to the abundance of European rabbits, *Oryctolagus cuniculus*?** *Wildlife Research* **41(2)**: 95-105 <http://dx.doi.org/10.1071/WR14033>

McGinness H. et al. (2014) **Floodplain amphibian abundance: responses to flooding and habitat type in Barmah Forest, Murray River, Australia.** *Wildlife Research* **41(2)**: 149-162 <http://dx.doi.org/10.1071/WR13224>

Stokehold D. et al (2014) **Factors influencing occurrence of a freshwater turtle in an urban landscape: a resilient species?** *Wildlife Research* **41(2)**: 163-171 <http://dx.doi.org/10.1071/WR13205>

## 2014 Ecological Consultants Association Conference August 8<sup>th</sup>

*'Ecological impact assessment - what is acceptable loss; and can consent conditions really assist as appropriate mitigation measures'*

The 2014 ECA conference was held at Noahs on the Beach in Newcastle. When not being distracted by the great view out the window, we heard from a variety of speakers, presenting on a wide range of topics. Sharon Molloy began the day discussing OEH's regional assessment project on ecological loss. Dr Stephen Ambrose used a case study involving Migratory Shorebirds to discuss 'Advocacy and Impartiality: examining the roles of Ecological Consultants'. Scott Duncan presented on Bio-certification within Wyong Shire and Robbie Economos presented the Lake Macquarie Regional Sustainability Planning Project. After a successful AGM and delicious lunch Peggy O'Donnell presented an example of Aquatic habitat creation as environmental mitigation. Matt Bell informed delegates of a Squirrel Glider case study in Great Lakes Council and Sarah Warner discussed impact assessment and acceptable loss for large forest owls. John Young finished the day with a presentation on the rediscovery of the Night Parrot and surveying for forest owls.

*Next years conference will be in Sydney, with a tentative date of the 31st of July*

## Recent Book Releases

Information Source: CSIRO Publishing  
Website <http://www.publish.csiro.au>

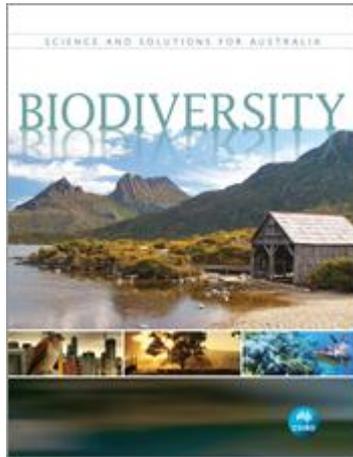
**Title:** Biodiversity: Science and Solutions for Australia

**Author:** Ed. S. Morton, A. Sheppard and M. Lonsdale

**No. Pages:**232

**Publisher:** CSIRO Publishing

**Date:** June 2014



**Title:** Sustainable Futures: Linking population, resources and the environment

**Author:** Ed. J Goldie and K. Betts

**RRP:** \$39.95

**No. Pages:**224

**Publisher:** CSIRO Publishing

**Date:** December 2014

**Title:** The World of Birds

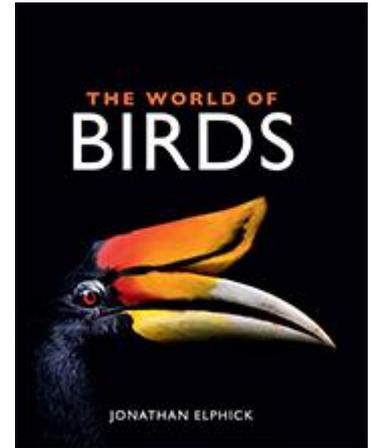
**Author:** Jonathan Elphick

**RRP:** \$89.95

**No. Pages:**612

**Publisher:** CSIRO Publishing

**Date:** September 2014



**Title:** Soils for Landscape Development: Selection, Specification and Validation

**Author:** S. Leake and E. Haegi

**RRP:** \$69.95

**No. Pages:**192

**Publisher:** CSIRO Publishing

**Date:** June 2014

**Title:** Carnivores of Australia: Past, Present and Future

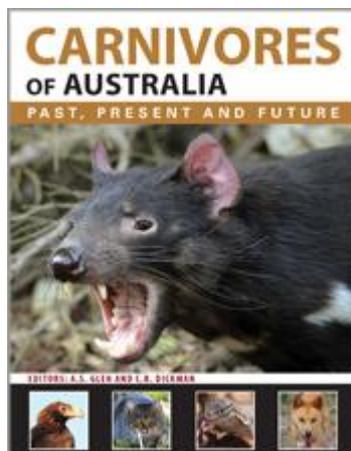
**Author:** A. Glen and C. Dickman

**RRP:** \$89.95

**No. Pages:**448

**Publisher:** CSIRO Publishing

**Date:** November 2014



**Title:** Camera Trapping: Wildlife Management and Research

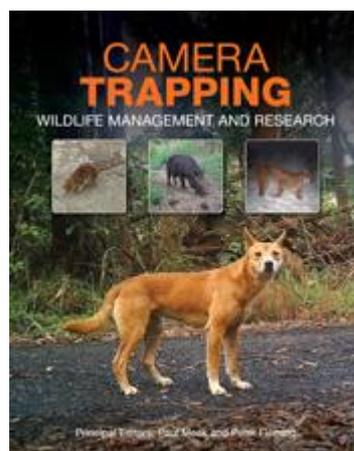
**Author:** Ed. P. Meek et al,

**RRP:** \$89.95

**No. Pages:**392

**Publisher:** CSIRO Publishing

**Date:** November 2014



## PHOTO COMPETITION

*Congratulations!* to **Phil Cameron** for winning the last photo competition with his photograph featured on the front cover of a Waratah.

Thank you to everyone who entered our photo competition. All entries have been included in the ECA Photo Gallery on the back cover and central pages of the newsletter.

*Email your favourite flora or fauna photo to [admin@ecansw.org.au](mailto:admin@ecansw.org.au) to enter a competition and have your photo on the cover of the next ECA newsletter. Win your choice of one year free membership or free entry into the next ECA annual conference. The winner will be selected by the ECA council. Runners up will be printed in the photo gallery*

*Photos entered in the competition may also be used on the ECA website*

# Wildlife Schools 2014

Practical ecology training courses, delivered by experts



Wildlife Schools are specialist training courses for environmental practitioners who need to develop their skills in the survey, identification and management of our flora and fauna.

At our courses there is an emphasis on time spent in the field, offering a rare opportunity to visit habitats and gain practical experience.

Course conveners Dr Frank Lemckert and Dr Rod Kavanagh have a wealth of knowledge to share, with additional presentations and demonstrations from other recognised experts with decades of experience.

- Learn from and be guided by recognised experts in Australian wildlife
- Experience habitats and a wide range of species first hand
- Build your confidence in practical ecology
- Gain a better understanding of biodiversity and environmental assessment
- See how environmental compliance is practically applied in the field

## REGISTRATION NOW OPEN

### General Survey and Identification Training Course

27-29 October, Crommelin Field Station, Pearl Beach NSW  
67.5 CPD points for Certified Environmental Practitioners (CENVP)

#### Registration enquiries:

Deretta Brown  
0488 774 107  
dbrown@niche-eh.com

#### General course enquiries:

Dr Frank Lemckert  
0425 249 026  
flemckert@niche-eh.com

Endorsed by



CPD points



Organised by



[www.wildliveschools.com.au](http://www.wildliveschools.com.au)

**FOR SALE / WANTED**

If you have 2nd hand ecological equipment that you would like to sell or would like to purchase you can place an ad in this newsletter. Free for members or \$40 for non-members.

Contact [admin@ecansw.org.au](mailto:admin@ecansw.org.au).



Environmental Insurance Specialists

Professional Indemnity & Business Insurance  
for Ecological Consultants.

Call Anthony Saunders or Hamish Robinson

Phone (02) 9949 1155 or 1300 799 950

Mackellar Insurance Brokers License 243531  
Suite 9, 67 Wanganella Street, (PO BOX 216)  
BALGOWLAH NSW 2093

*"Non-ECA promotional material presented in the ECA Newsletter does not necessarily represent the views of the ECA or its members."*

# GETTING MORE FROM YOUR BAT SURVEYS

Anna McConville  
Echo Ecology

Insectivorous bats are often required to be surveyed as part of the ecological impact assessment process in NSW. Biodiversity survey guidelines for NSW recommend using bat detectors to record ultrasonic bat calls combined with harp trapping to sample insectivorous bats (DEC 2004). Bat detectors are easy to set, are non-invasive, and can be left in place to sample for long periods. For these reasons, they are regularly deployed by ecological consultants that are undertaking fauna surveys. But did you know that we can do much more with the recordings from bat detectors than just species records? We can also use bat detectors to look at bat activity, which allows us to compare or rank different habitats, sites, or treatments in an experimental setting.

There are lots of different bat detectors on the market that record the high frequency echolocation calls made by insectivorous bats. In Australia, people are most familiar with the Anabat range of bat detectors (Titley Electronics, Australia). More recently, Song Meters (Wildlife Acoustics, USA) have become popular due to their low cost and ability to record acoustics (frogs, birds, etc) at the same time as ultrasonics. Regardless of the model, the bat call recordings made by the bat detectors can be plotted as time vs frequency graphs, and the species identified using regional bat call identification guides (e.g. Pennay, Law *et al.* 2004). While bat detectors can't be used to determine abundance (as individuals cannot be distinguished), the number of call files attributed to each species can be tallied to create an index of activity. We can then use these activity levels to address a range of different questions in a similar manner to the way we use abundance for other species.

But there are a few things that we need to do in order to ensure that we are collecting data in a way that we can use in an experimental setting. So I thought it

might be useful to run through the basics of how to use activity levels.

## How do we do this?

I have described my simple approach to using bat activity levels in experimental design. There are a lot of other things that you could fine tune depending on your questions and what you want to achieve; but this is what I think is achievable in a standard fauna survey without too much additional time.

### Step 1 - Calibrate your bat detectors

When comparing bat activity between different experimental units, it is really important that your bat detectors are sampling the same amount of airspace. Unfortunately, as bat detectors age, their sensitivity usually declines as a result of microphone damage and general wear and tear. So the actual sensitivity level will vary among detectors and you will need to calibrate them prior to undertaking a comparative study.

See Appendix 1 for how to calibrate multiple bat detectors.

### Step 2 - What are your questions?

This is the stage that shapes your study and you need to decide what the purpose of your study is. You need to come up with a question or prediction that you can base your study on.

For example, a common question for consultants may be: how does bat activity relate to habitat type? This may be for an entire bat community (total activity), a foraging guild, or a single threatened species. By knowing what habitat types are used the most by insectivorous bats, or particular threatened insectivorous bats, we can better understand the potential impacts of development proposals. For long term monitoring studies, we may also be interested in how bat activity changes over time in response to management actions or impacts.

### Step 3 - Plan your sites

The questions that you are interested in exploring will

drive this stage. There are also a number of practical considerations that you will need to think about, including the number of bat detectors you have and site access. But don't worry: this planning process doesn't have to take up too much time, particularly if you have prepared a similar study previously.

If we were looking to compare bat activity among different habitat types, we would begin by stratifying the site by broad vegetation type that we think may be relevant to bats. For example, we may divide a large site into cleared paddock; cleared paddock with scattered paddock trees; moderate condition dry sclerophyll forest (e.g. spotted gum/ironbark forest with large hollow-bearing trees); regrowth dry sclerophyll forest (e.g. spotted gum/ironbark forest which has been historically cleared and has few hollows); riparian forest (e.g. red gum forest); swamp forest (e.g. paperbark/swamp mahogany forest); and freshwater wetland (e.g. dam with open water and reeds).

We would then plan to set bat detectors in each of these habitat types, with replication (at least three different sample sites per habitat type). Ideally, you will randomly select sample sites within these stratification units. However, random sampling is not always possible due to site access and other issues. So if you can't randomly sample, then try to develop some site selection rules to make the site selection process as objective as possible.

This process is something that you would probably be doing anyway to meet survey guidelines.

#### Step 4 - Standardise sampling

When comparing the results from different sites, we want to make sure that we are detecting actual differences among our treatments, and not some other variation. So you need to make sure that you are consistent in the way you sample among sites.

You would have already calibrated your bat detectors, so you are one step closer to meeting this. Here are some suggestions on the types of things to standardise among your sites:

- Weatherproofing and microphone extension cables;
- Height of microphone;
- Angle of microphone;
- Division ratio;
- Amount of clutter. If you are sampling open habitats as some of your categories, make sure that any detectors placed in forests are aimed along tracks or placed in more open spaces so that vegetation doesn't interfere with detection;
- Duration of sampling (including provision for adequate batteries);
- Use only the same brand of bat detector for each study;
- Weather. Avoid or discard recordings from nights that have high wind, high rain, full moon or are unusually cold, as these have been found to be related to low bat activity in some studies.

Most studies will not have enough bat detectors to be able to sample every site on the same nights. This is fine as long as you try to minimise the amount of time between samples. Avoid spreading sampling over a number of different seasons (unless that relates to your question) as bats will be doing different things at different times of year. Also, if you are aiming to compare bat activity among different habitat types, try to sample a mix of different habitat types at any one time. For example, you don't want to sample all of your freshwater wetlands on the first two nights, then all of your dry sclerophyll forest on the next two nights.

Consider adding an extra site or two than you think you need to compensate for the 'field work factor', where nothing ever goes to plan. Forgetting to turn on bat detectors, battery failure, memory card corruption, etc, are all things that can mean you end up with data from fewer sites than you originally plan.

The most time-consuming part of standardising your sampling is calibrating your bat detectors. However, you usually only need to do this once a year so it doesn't actually add much time to each project. Also, I set my bat detectors in the same way for each survey, so this step is not something that I need to think about in detail before each study. Think about it once; put the process in place; and move on.

Step 5 - collect data

Go forth and set your bat detectors following your plan. This can usually be combined with other fauna survey work. It is a good idea to pay careful attention to file naming and backups so that you don't lose your data.

Step 6 - interpret the results

Once you have downloaded your data and sent it off for identification, you will end up with a tally of the number of bat call files (also known as passes), and can

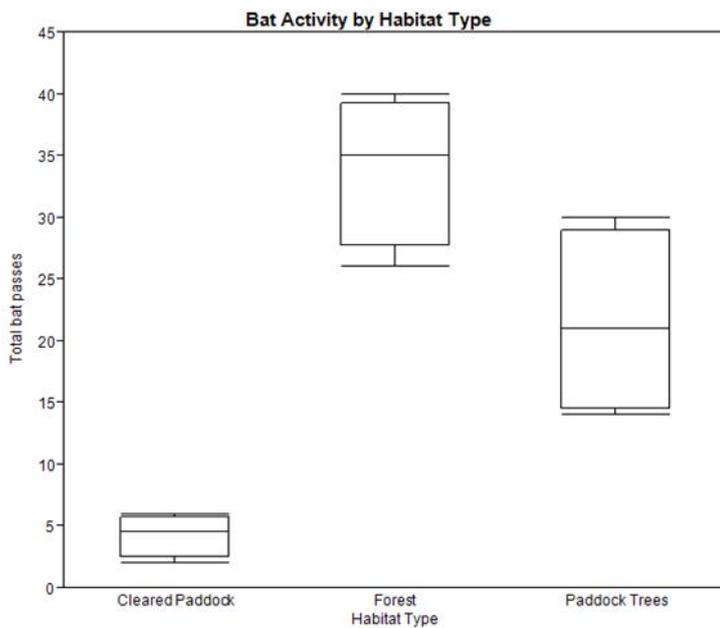
create a results table similar to that provided in Table 1.

Once you have your bat activity levels, you can then do some statistics to compare activity levels between sites. How complicated you go with your statistics is up to you, and often depends on your experience and confidence. It is always a good idea to do some exploratory graphing to get an idea of how your different categories match up. Box plots are a good way of doing this, and they often let you know at a glance if you are getting some differences between your categories or you have something strange going on with your data (Figure 1).

**Table 1: Example bat activity (number of passes) per site**

Site	Type	<i>Chalinolobus gouldii</i>	<i>Chalinolobus morio</i>	<i>Miniopterus australis</i>	<i>Mormopterus (Micronomus) norfolkensis</i>	<i>Mormopterus (Ozimops) ridei</i>	<i>Rhinolophus megaphyllus</i>	<i>Scotorepens balstoni</i>	<i>Vespardelus vulturinus</i>	TOTAL
1	Cleared Paddock	2	0	1	0	2	0	0	0	5
2	Cleared Paddock	1	0	0	1	1	0	1	0	4
3	Cleared Paddock	3	1	1	0	1	0	0	0	6
4	Cleared Paddock	0	1	0	0	0	0	0	1	2
5	Paddock Trees	5	2	2	3	4	0	0	0	16
6	Paddock Trees	7	4	1	6	5	0	5	2	30
7	Paddock Trees	3	3	2	10	1	0	2	5	26
8	Paddock Trees	4	1	2	3	3	0	1	0	14
9	Forest	5	3	4	1	4	7	3	10	37
10	Forest	8	2	3	0	2	5	4	2	26
11	Forest	6	1	10	1	3	4	1	7	33
12	Forest	6	3	4	1	3	5	8	10	40

**Figure 1: Example box plot of bat activity (total number of passes) by habitat type category**



You can tally total bat activity between habitat types by pooling the total number of bat passes, regardless of species. You can look at individual species if you have enough passes or you could group your species into foraging guilds (e.g. open-space, edge-space and clutter-space foragers). This may be particularly useful for wind farm projects where you may be more interested in high-flying species (open-space foragers), like the White-striped Free-tailed Bat (*Austronomus australis*). You still get presence data from your study and can continue to report and plot your threatened species data as usual.

This is by no means an exhaustive list of all the experimental considerations, questions and analyses techniques that you can do with bat activity data; but I hope that it inspires you to look a bit deeper and to get more from your next bat survey.

## Appendix 1 - How to calibrate your bat detectors

As Anabat units (Titley Electronics, Australia) have been the most common bat detector used in NSW, I have described a calibration process for these units. But you can apply the principles to other bat detectors and many manufacturers offer calibration packages and instructions.

### 1. Purchase a ready-made calibration package

If you want to make this super easy you can purchase

an Anabat Equalizer from Titley Electronics, Australia for around \$800. This combines a bat chirper, which is a device that emits a high frequency tone, with a jig to hold your bat detector in place; and comes with software that automates the process.

If you don't have a budget that covers this, then read on for a relatively simple calibration process that you can do without purchasing any additional equipment.

### 2. Do it yourself

You can do this for free with your existing equipment and a little bit of trial and error. A bat chirper is a small device that emits a constant high frequency tone and is a handy addition to the calibration process. They are available for purchase from a number of different manufacturers. Don't worry if you don't have one. You can still calibrate your detectors by using the high frequency noise your fingers make when rubbed together.

#### *Step 1 - inspect your bat detectors*

Before you start, it is a good idea to assess the condition of your bat detectors. Inspect the microphone for damage such as discoloration, peeling or rusting of the metal filament. Such damage will substantially affect the performance and sensitivity of your bat detector.

Also keep in mind that Anabat SD1 and SD2 units have internal batteries (different to the ones that you put in for each survey) that require professional replacement every couple of years. It is really frustrating to go to the effort of setting out bat detectors to find that they fail due to internal batteries. So if you haven't got the internal batteries replaced in a while or the microphone looks in bad condition, send the bat detector away for a service before you calibrate.

#### *Step 2 - find a suitable location*

Temperature, wind, humidity and vegetation can all influence the volume of airspace that your bat detector is able to sample. So we want to minimise the variation in conditions during the testing of each bat detector. A

large open room like a warehouse or large garage is perfect. You can also do this outside, but just make sure you have a nice open space and it is not windy. I have used a long and wide driveway before. You should also calibrate all the detectors on the same day to minimise the temperature and humidity differences.

### *Step 3 - make your own jig*

We want to end up with a bat detector at one end of the open space and a bat chirper (or your fingers) at the other end of the open space. Initially, we will have to adjust the distance between the two. But once we have settled on the distance, we will need something to place the bat detector (and chirper) on so that we can ensure that each bat detector will lie in exactly the same position and direction. I like to get the detector up off the ground, so I use two milk crates stacked on top of each other at either end. I refer to these throughout as the detector stand and the chirper stand.

I then securely attach a piece of plywood to the top of the detector stand onto which I draw an Anabat outline. I do the same for the chirper stand. The chirper outline can just be a circle for you to place your hand if you aren't using a chirper. You will test one detector at a time, so you only need one setup.

### *Step 4 - find the least sensitive detector and set the distance*

At this stage you want to get a rough idea of which detector is the least sensitive. So you will need to have all the bat detectors with you that you wish to calibrate.

Set your crate stands apart (maybe 5 m apart initially) and place the first bat detector on one end. Turn on the bat detector.

Most of the Anabat models have a sensitivity dial on the front of the unit to allow easy adjustment. Turn the sensitivity dial of the bat detector up until you hear lots of static noise and then turn it back down a little until the static noise disappears.

Place the bat chirper on top of the chirper stand and turn it on. If you don't have a bat chirper, then one

person needs to stand at the chirper stand end and rub their fingers together. You can rest your hand on top of the crate to keep the position relatively constant. You don't have to line up the detector perfectly at this stage as this is just a rough measurement.

You should now be able to hear the bat chirper or fingers signal on the bat detector. If not, move the bat detector closer to the chirper stand until you hear it. Once you do hear the noise, move the bat detector back away from the chirper stand until you reach a point just before you lose the signal. This may require a little bit of back and forth adjustment.

Once you have found that point, set the detector stand at this location. Select the next bat detector, turn the sensitivity dial up until you hear the static noise (not the bat chirper or fingers noise), and then turn it back down a little until the static disappears. Place it on the detector stand and see if you can hear the bat chirper or fingers signal. If you can, then move onto the next detector as this is not the least sensitive detector.

Repeat this process until you find a detector that cannot detect the chirper signal. Once you find a detector that can't detect the chirper or fingers signal, move the detector stand closer to the chirper stand until you can just hear the signal. Keep repeating this process until you go through all of your bat detectors. You will end up with one detector that needs to be closer to the chirper than all of the others when set to its maximum sensitivity level. This is your least sensitive bat detector.

You will note that the maximum sensitivity number on the dial (above which you get the static noise) will be different among detectors. This is perfectly normal and this is the reason that we are calibrating them in the first place!

Now that you know which is the least sensitive detector, you can set your calibration distance. Here you want to be very careful about the placement of the detector and chirper stands. As Anabat detectors are directional, you need to ensure that each subsequent detector is aiming at exactly the same angle (straight on) towards the chirper stand.

The detector and chirper stands need to be at a distance apart where you can just hear the signal on the least sensitive detector. This is where a windy day can play havoc with calibration and should be avoided. Have a bit of a play around and get this position just right.

Once you have, make sure that your stands are secure and nothing is going to move. If you are using your fingers to produce the signal, then try to do this as consistently as possible and from the same point on the chirper stand.

#### *Step 5 - calibrate the detectors*

Your least sensitive detector should be set on the highest sensitivity level on the front dial just below the level at which you hear the static noise. This will be the sensitivity setting you will always use for this detector during surveys. So you need to mark this setting. I use a lead pencil to make a mark on the sensitivity dial of the bat detector itself. The pencil mark lasts well, but is easy enough to clean off next year. If you have previous marks, then remove these prior to calibrating.

Once you have done this you will calibrate all of the other bat detectors to this least sensitive bat detector. You no longer have to move any equipment towards or away from each other. You will only need to adjust the sensitivity dial.

Select the next bat detector and place it on the detector stand. As the first detector was the least sensitive, all of the other detectors should be able to detect the chirper or fingers signal as soon as you place them on the detector stand. If they can't, you will need to go back to Step 4 and repeat.

Once the detector is in place on the detector stand, reduce the sensitivity level on the dial until you can only just hear the chirper signal. This will be the final sensitivity level for this detector. Make a pencil mark on the sensitivity dial. Repeat this process for all of the remaining bat detectors. You will end up with your least sensitive detector being set at something like 9 and your most sensitive detector set at something like 6. If your least sensitive detector is really terrible

compared to your others, consider sending it off for a service before continuing with the calibration.

So now you have a suite of bat detectors, each with a little mark on the sensitivity dial. All you have to do each time you set your bat detector out for a survey is to set the sensitivity to the mark on the dial and you will be able to compare results among detectors. Once you have calibrated your bat detectors they are usually fine for the remainder of that field survey season.

If you get new microphones, new detectors or experience bad weather conditions that may have damaged your microphones, then you should calibrate again. This sounds like a long process, but it doesn't take more than an hour and is much quicker once you get the hang of it.

#### *A couple of notes*

Don't swap microphones between bat detectors after you have calibrated them. This will change the sensitivity level of each unit. If you do have to change microphones or purchase a new one, you will need to repeat the calibration process.

A neat tool to illustrate how bat detector sensitivity settings, temperature and humidity all affect the amount of air space volume you sample is AnaVolumes, which is available free from Chris Corben's website ([www.hoarybat.com](http://www.hoarybat.com)).

#### **References**

DEC (2004) *Threatened Biodiversity Survey and Assessment: Guidelines for Developments and Activities (working draft)*. NSW Department of Environment and Conservation, Hurstville, NSW.

Pennay, M., Law, B., and Reinhold, L. (2004) **Bat calls of NSW, region based guide to the echolocation calls of Microchiropteran bats**. NSW Department of Environment and Conservation, Hurstville.

## CREATING ROOSTING HABITAT ALTERNATIVES FOR BREEDING MYOTIS

Veronica Silver and Anna Lloyd  
GeoLink

During recent field investigations undertaken as part of a Review of Environmental Factors for Roads and Maritime Service for road widening and culvert repairs, GeoLINK proposed a new approach to creating alternative roosting habitat for breeding Myotis. Large-footed Myotis (*Myotis macropus*) were found roosting within a 1200mm three-cell concrete pipe culvert in northern NSW. The Large-footed Myotis is listed as Vulnerable within Schedule 2 of the *Threatened Species Conservation Act 1995*.

A Bat Management Plan, which prescribed provision of alternative habitat and monitoring post construction, was prepared to provide a procedure to be followed prior to and during construction works to minimise potential impacts to microbats.

Initially, planning involved a two-staged habitat replacement procedure. Seven bat boxes were hung near the culvert outlet from branches of a large Small-leaved Fig (*Ficus obliqua* var. *obliqua*) that were overhanging the waterway (refer to **Plates 1** and **2**). These bat boxes were installed approximately nine months prior to works and were to be temporary measures to provide immediate habitat whilst the old culvert was dismantled and a new culvert with permanent habitat constructed. Bat boxes are known to provide suitable roosting habitat for Myotis and we have been using bat boxes successfully on other projects. The down side of the bat boxes, particularly when used in trees, is that they are open to the elements, requiring on-going maintenance, and also the readiness with which they are used by breeding females is unclear.

In response to the need to provide permanent and appropriate breeding habitat, various options were assessed to simulate the 'cave-like' or earthen recesses that were available in and utilised by breeding females in the old culvert.

Initially "recessed chambers" and "baffles" were designed to provide habitat within the area between the top of the culvert and the road surface above (refer to **Figure 1**). The aim was to reduce the potential impacts of flood events and log debris damaging the habitat, but to also attempt to re-create similar



**Plate 1 – Nest boxes in Small-leaved Fig**



**Plate 2 –Myotis within 2nd lowest nest box**

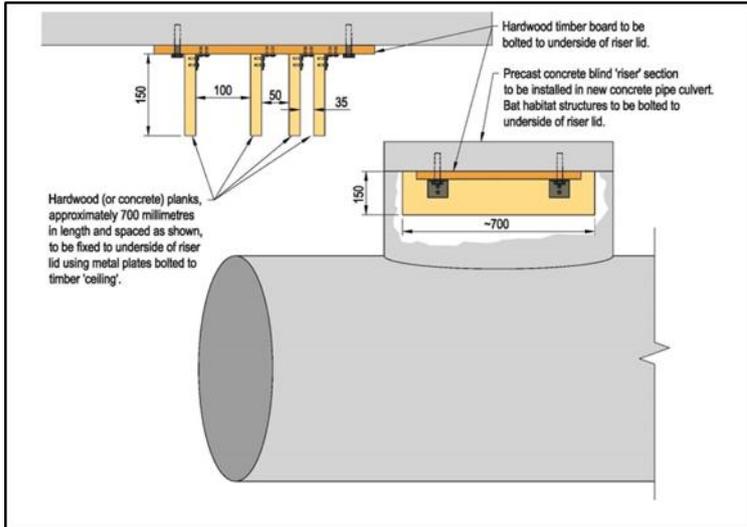
microclimate conditions as are provided for bats in old structures where cracks in concrete and fall-out of soil creates earthen domed cavities for roosting. These were then to be mounted to the top of the culvert.

In consultation with engineers, these ideas were modified to comprise of standard bat boxes (instead of baffles) installed within two 'chambers' which were actually pre-manufactured 'man holes' (refer to **Plates 3-6**). The man holes were covered with a standard concrete lid, and soil and road material covered over the new culvert as per the original plans.

It was found that the temporary bat boxes installed in the Small-leaved Fig were utilised and provided important temporary and immediate habitat for newly evicted Myotis. However not all of the excluded bats used the tree-mounted bat boxes, and only the three lowest nest boxes were used. Monitoring has been undertaken from December 2012 to April 2014, and the boxes continue to be used after the permanent habitat became available.

Breeding activity has also been noted in the tree-

mounted bat boxes, therefore as a result of their continued use, the 'temporary' habitat will be left in situ.



**Figure 1. Baffles within a man hole were proposed but a more practical solution of pre-fabricated nest boxes were installed**

Four bat boxes were installed within each man hole. Although evidence of use in the form of staining and guano has been observed in the man hole closest to the inlet (north), microbats have only ever been observed in the second man hole. Since monitoring the permanent alternative habitat provided within the culvert, bats have used the man holes and bat boxes for roosting and breeding. Whilst numbers have not returned to the numbers prior to removal of the old culvert in June 2013, they are increasing and are now approximately 55% of the original number of bats contained within the culvert. Monitoring will continue to determine if numbers return to similar levels.

Without the use of radio-telemetry or a permanent bat marking (e.g. banding), it is not possible to determine if the bats in the new culvert contain individuals that utilised the old culvert.

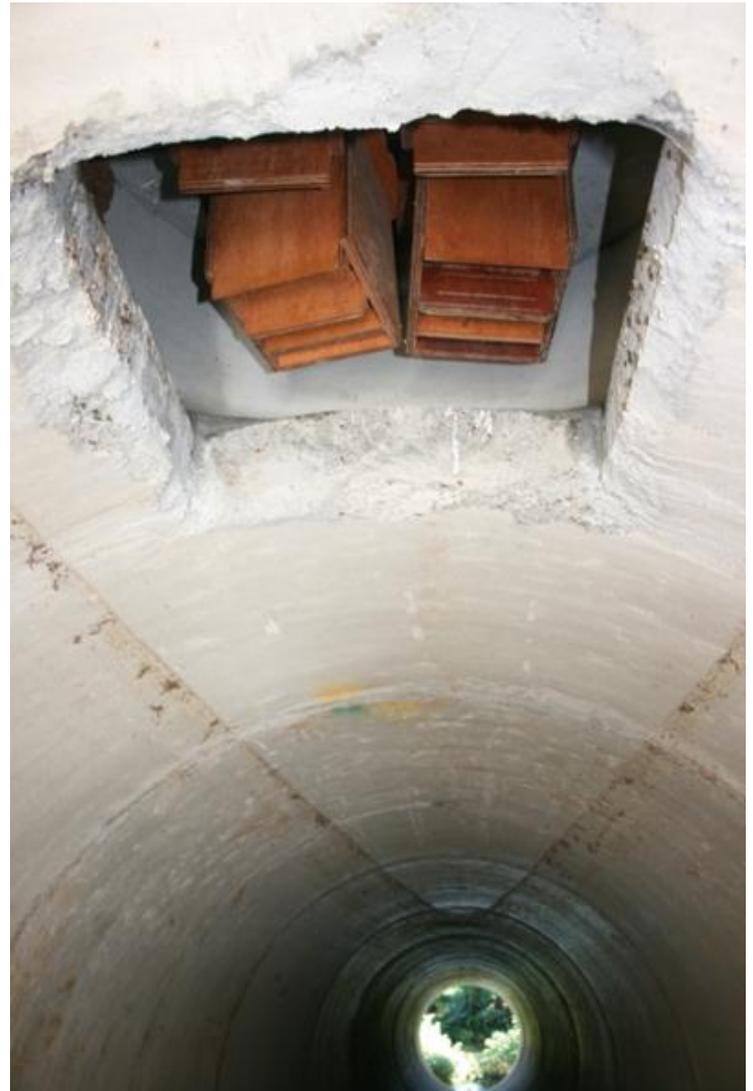
In addition to these alternate types of habitat, culvert lift points were also utilised by microbats. Culvert lift points are small cavities in the concrete, cut for ease of lifting pipes during installation (refer to **Plates 7** and



**Plate 3 – Approximately ten Myotis within nest box of chamber 2**



**Plate 4 – Approximately ten Myotis within nest box of chamber 2**



**Plate 5 – Bat boxes within chambers (view south to outlet)**



**Plate 6 – Bat boxes within chambers (view north to inlet)**

8). These cavities were not deliberately provided as habitat, however in suitable locations, the culvert lift points have been used following completion of works. The suitable locations at the subject site were in the middle (where it was darker), and near the outlet (closer to a permanent source of water). I have seen these culvert lift points filled in with grout/concrete/plastic plug following completion of works at other sites, however now I would recommend that these are not filled in as they provide suitable habitat for microbats. Further to this, I would also suggest installing a concrete box/chamber above the lift holes, and if possible, do not grout culvert joints and lay pipes with maximum jointing gap (as specified by manufacturer).

Design modifications and lessons learnt include:

- The chambers (or man holes) need to be installed in the most suitable location for the microbats (if the road design permits). In this case, it would have been at the outlet end which had more shade and water.
- Ensure microbats have adequate access to all nest boxes located within the chambers by making the

chambers slightly bigger and having longer length landing pads.

- Consider an alternate design with a lattice structure to provide greater carrying capacity.
- Install a central post extending down with gaps radiating out to provide greater yield potential.
- A deflector could be installed on the upstream side of the manhole to guide flood debris down below it (if regular flooding is likely).



**Plate 7 – Myotis within culvert lift point**



**Plate 8 – Culvert lift point**

Although the Myotis habitat in the form of cracks of the failing culvert were removed, breeding Myotis activity has been observed in two monitoring events post construction within the nest boxes in the chambers and the nest boxes on the fig tree at the outlet. This project shows that recesses can be introduced into culverts using pre-fabricated man holes, and that these can be fitted with bat furniture to provide breeding habitat for Myotis.

This project also capitalised on the availability of suitably structured trees overhanging a waterway to provide 'temporary' habitat whilst construction was underway.

We were also grateful that NSW Roads and Maritime Service was open to suggestions of provision of different types of alternative habitat, and see this program providing valuable feedback to on-going and pro-active microbat management for RMS projects.



Plate 9 –Myotis within 2<sup>nd</sup> lowest nest box



Plate 10 – Appropriately named LARGE-FOOTED Myotis

## HOLIDAYING OR MOVING IN?: THE STORY OF THE MOST SOUTHERLY CONFIRMED RECORD OF NOISY PITTA (*PITTA VERSICOLOR*) AT MORUNNA POINT, WALLAGA LAKE, NSW FAR SOUTH COAST.

S. Sass<sup>1,2</sup>, A. Morrison<sup>3</sup>, & H. Warne<sup>4</sup>

<sup>1</sup> EnviroKey, P.O.Box 7231, Tathra NSW 2550

<sup>2</sup> Institute for Land, Water and Society, Charles Sturt University, Thurgoona NSW 2640

<sup>3</sup> Bega Valley Shire Council, PO Box 492, Bega NSW 2550

<sup>4</sup> 22 Lloyd Street, South Pambula NSW 2549

Email [steve@envirokey.com.au](mailto:steve@envirokey.com.au)

### First reported sighting

On the 12<sup>th</sup> July 2014, Mr. Christopher Hemmingsen posted a photograph in the Facebook group 'Birds of Oz' that he had taken of a bird 'near Bermagui' and asked for assistance with identification. Despite the bird depicted in the image being well distant and in low light, there was little doubt as to its identification. It was a Noisy Pitta (*Pitta versicolor*). One author (SS) messaged Christopher to request clarification on the location in which he responded with precise details of the location which was Morunna Point (36°22'20.20"S; 150°4'42.29"E). Christopher stated on Eremaea Birdlines that two individuals were observed. While Higgins *et. al* (2001) note unconfirmed historical reports of Noisy Pitta at Mallacoota, Victoria (around 130 kilometres south of Morunna Point), this short note documents the most southerly confirmed record of Noisy Pitta in Australia.

### Our sighting

On 14<sup>th</sup> July 2014, we arrived onsite at around 6.35am and although still very low light, we heard the distinct hopping of a Pitta moving through the leaf litter. We retreated some 50m until light conditions improved, and at around 7am, we moved back to the same position. A single Noisy Pitta was observed foraging amongst a dense stand of regrowth Sweet Pittosporum

(*Pittosporum undulatum*) (Plate 1). No further Noisy Pitta were observed.

**Plate 1: Noisy Pitta observed at Morunna Point, Wallaga Lake, NSW far south coast on the 14<sup>th</sup> July 2014 (Photographs by Steve Sass, EnviroKey).**



### Known Distribution

Higgins *et. al* (2001) report that the Noisy Pitta has a widespread distribution from islands in the Torres Strait to the Hunter region and mostly east of the Great Dividing Range. In NSW, they are regularly recorded north of Taree and irregularly recorded as far south as Sydney and the Illawarra with relatively recent records in Kiama (1992) and Jamberoo Pass (1990).

Post Higgins *et al* (2001) is the record of a single individual (juvenile) at nearby Akolele in 2002 (Eurobodalla Natural History Society: Julie Morgan, recorder - from Eremaea Birdlines) less than 2 kilometres north of Morunna Point. A search of the Atlas of NSW Wildlife confirms that the majority of Noisy Pitta records are north of Sydney (Figure 1).

### Morunna Point Record

Various birdwatchers have visited the Morunna Point site in an attempt to observe the most southerly Noisy Pitta in Australia. Some, including the initial observation by Mr. Christopher Hemmingsen, noted at least two birds. A posting on Eremaea (14/07) also confirmed at least two Noisy Pitta (Mandy Anderson and Richard Nipperess) with one individual observed and a second bird heard calling from several metres distant in the dense vegetation. They also note the movement of 'a possible third bird'. We could only confirm the presence of one individual.

Given this information, it can be stated that there are likely to be at least two individual Noisy Pitta present. There has been some discussion elsewhere as to whether the individuals present at Morunna Point are aviary escapees, which is noted against the Wollongong observations by Higgins *et. al* (2001). I have contacted several aviculturists who breed Noisy Pitta in captivity in NSW, and they have noted that the species is extremely rare in captivity, held by individuals under an advanced bird keepers license, and none are aware of any in captivity in the far south coast region. Further, I have made personal contact with a number of aviculturists in the district and these conversations confirm that Noisy Pitta is not currently held in any south coast aviaries. Given this, it can be stated with an extremely high level of confidence that the individuals at Morunna Point are not aviary escapees.

### Holidaying or Moving In?

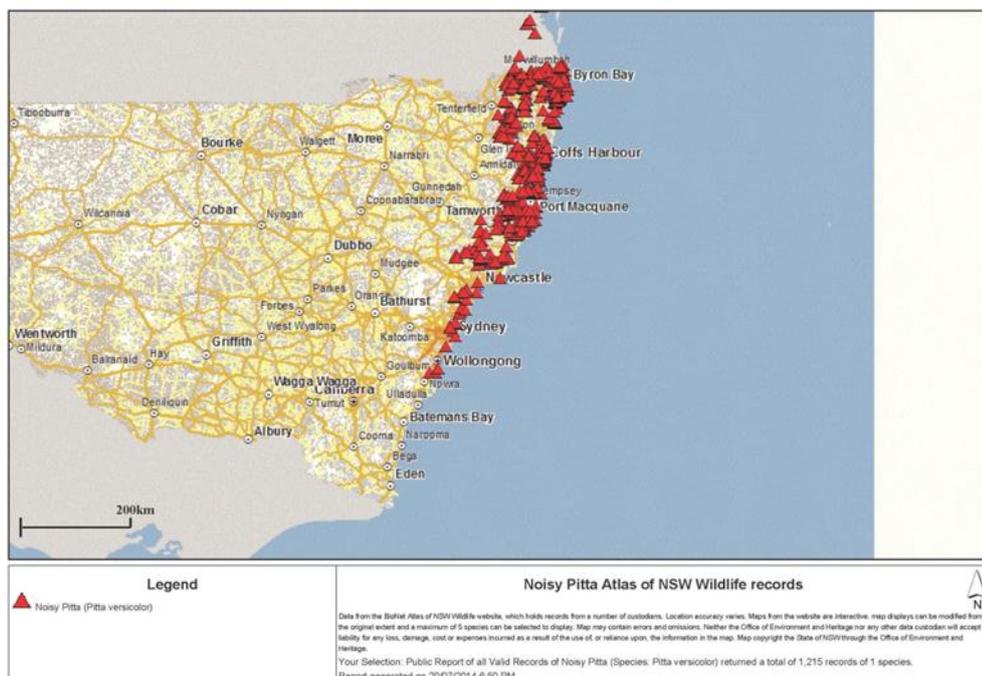
Noisy Pitta are a relatively conspicuous bird having a distinctive call and the species is regularly recorded across its range. The Far South Coast Birdwatchers Group comprises a group of well-experienced birdwatchers, and frequently survey the Morunna Point site as part of their regular outings. It is highly probable that if Noisy Pitta was present for an extended period of time, that any individuals would have been detected by members of this group.

Although movement ability is not well known, Noisy Pitta is considered nomadic, or at least, partially

nomadic. Noisy Pitta is now regularly recorded at a number of locations in and around Sydney and these constant sightings may suggest a more permanent shift in the southern extent of distribution. Chambers *et. al* (2005) maintain that these southern range extensions are consistent with Climate Change. Roberts (2003) confirms changes in movement patterns in south-east Queensland. Noisy Pitta were known to move to the warmer lowland regions during winter, returning to the mountains to breed. However, they now spend the entire year in the mountain forests where minimum temperatures have warmed over the past half century. Noisy Pitta at Morunna Point, and indeed the 2002 observation at nearby Akolele, may be part of a southerly shift in distribution as suggested by Chambers *et. al* (2005). The habitat at Morunna Point is consistent with the known habitat for Noisy Pitta in the north of their range being a closed canopy with sparse ground vegetation.

The authors cannot rule out the presence of Noisy Pitta elsewhere on the NSW far south coast. Nearby Mount Gulaga (less than 6 kilometres north west of Morunna Point) provides significant areas of rainforest habitat and may provide an important landscape feature in a southerly shift in distribution. Littoral rainforest and thickets of *Pittosporum* spp. are also likely to provide areas of potential habitat for Noisy Pitta on the NSW far south coast.

Holidaying or moving in? Given the information at hand, it is probable that a southerly shift in distribution is the most likely scenario in relation to the presence of Noisy Pitta on the NSW far south coast. The confirmation of a juvenile at Akolele in 2002 and a juvenile at Morunna Point suggests that Noisy Pitta may be breeding on the NSW far south coast. With regular reports of Noisy Pitta in the Sydney and Illawarra region, it is plausible that the known breeding limit may also be extending southward.



**Figure 1: Records of Noisy Pitta from the Atlas of NSW**

We hypothesise that Noisy Pitta sightings will become more frequent across the NSW far south coast. We recommend that regular surveys be conducted in areas of Littoral rainforest and thickets of *Pittosporum* spp. to monitor for any individuals and to confirm if Noisy Pitta are breeding in the region. Finally, the use of social media to identify populations of species outside of their known distribution should not be underestimated.

### Acknowledgements

We acknowledge and thank Mr. Christopher Hemmingsen for first reporting the presence of Noisy Pitta at Morunna Point in the Facebook group 'Birds of Oz'. We also thank Barbara Jones from the Far South Coast Birdwatchers for encouraging this short note. Finally, we thank the numerous aviculturists that we spoke with in relation to any captive populations of Noisy Pitta.

### References

Chambers, L. E; Hughes, L and Weston, M.A (2005) Climate Change and its impact on Australia's avifauna. *Emu* 2005, 105, 1-20.

Higgins, P. J, Peter, J.M & Steele, J.K (Eds.)(2001). **Handbook of Australian, New Zealand and Antarctic Birds. Volume 5: Tyrant-flycatchers to Chats.** Oxford University Press, Melbourne.

Roberts, G. (2003). Climate spins whole new web of life. In *'Weekend Australian'*. 25 October 2003, p. 2.

## TRAPS, TRANSECTS AND TECHNOLOGY: DETECTABILITY OF THE THREATENED ROSENBERG'S GOANNA (*VARANUS ROSENBERGI*)

S. Sass<sup>1,2</sup>, J. Murray<sup>3</sup> & K. Chesnut<sup>4</sup>

<sup>1</sup> EnviroKey, P.O.Box 7231, Tathra NSW 2550

<sup>2</sup> Institute for Land, Water and Society, Charles Sturt University, Thurgoona NSW 2640

Email [steve@envirokey.com.au](mailto:steve@envirokey.com.au)

<sup>3</sup> Biosis, 14/17-27 Power Avenue, Alexandria NSW 2015

<sup>4</sup> GHD, 15/133 Castlereagh Street, Sydney NSW 2000

### Introduction

The Rosenberg's Goanna *Varanus rosenbergi*, also known as the Heath Monitor, is a medium sized varanid that is currently thought to have a disjunct distribution across southern Australia (**Plate 1 & 2**). It is known to occur in the sandy soils of southern WA, SA and western Victoria; and in the Snowy mountains and mid-coast regions of NSW (Clemann *et al.* 2005; DECC 2007; Swan *et al.* 2004; Wilson and Swan 2013). An island population is also known from Kangaroo Island in South Australia (King and Green 1999).

In NSW, the species is listed as Vulnerable under the NSW *Threatened Species Conservation Act 1995* (OEH 2014b). Despite this, the ecology of the species has received little attention with the exception of the Kangaroo Island population which has been extensively studied (Green *et al.* 1991; King and Green 1999; Rismiller and McKelvey 2000). However, it is generally accepted that the species occurs within a variety of habitats including coastal heathlands, wet and dry sclerophyll forests, woodlands and mallee (Cogger 2000; Sass 2008; Swan and Watharow 2005; Vincent and Wilson 1999; Wilson and Swan 2013). Regardless of these generalisations, most observations of this species are based on opportunistic records rather than being the subject of detailed target surveys. Given this, there is a paucity of detailed information on

detectability of Rosenberg's Goanna in areas of occupied habitat.

Traditionally, Rosenberg's Goanna were targeted using baited cage traps or opportunistically by traversing suitable habitat on foot or vehicle. However, with the ongoing development of motion-activated cameras and other methodologies available to researchers, a comparison of potentially suitable methods was warranted. This manuscript provides a comparison of three potential detection techniques pooled from a number of previous surveys conducted by the authors; driving transects, unbaited funnel traps and motion-activated cameras in areas of habitat where Rosenberg's Goanna are known.



**Plate 1: Rosenberg's Goanna in defensive mode (photo courtesy and copyright Nick Clemman.**

### Methodologies

#### *Driving Transects*

Driving transects were undertaken along vehicular tracks in two areas where Rosenberg's Goanna are known. During each study, transects were undertaken from a slow moving vehicle with two observers (including the driver) visually scanning for Rosenberg's Goanna.

In a study by one of the authors in Summer 2008 (SS), 2178 kilometres of driving transects were completed in areas of habitat where Rosenberg's Goanna had been previously recorded (Bionet). Driving transects in that survey were completed driving at around 40km/h. These driving transects resulted in the detection of

three individuals: all road-killed specimens, along Main Road 92 between Sassafras and Nowra ( $\mu = 1$  animal/726 km) (Sass 2008).

In a Biodiversity Study conducted across the Liverpool Military Area in Spring 2010 and Autumn 2011, driving transects were undertaken along vehicular tracks (fire trails). A total of 326 kilometres were completed across known habitat which was confirmed by previous records (OEH 2014a), as well as one individual being trapped during the survey period. No individual Rosenberg's Goanna were detected during the driving transects ( $\mu = 0$  animals/326 km).



**Plate 2: Close-up of Rosenberg's Goanna showing distinctive lower lip markings (Photo courtesy and copyright Stuart Cooney).**

#### ***Motion-activated Infrared Cameras***

Motion-activated Infrared cameras are known for their efficiency in detecting fauna species without the need to set traditional traps (Claridge *et al.* 2004). During a study in Autumn 2013 in northern Sydney at a location where Rosenberg's Goanna are frequently observed, three Reconyx PC900 HyperFire Professional High Output motion-activated infrared cameras were activated for a period of five days. Each camera was at least 200 metres apart and aimed at a single bait attractant. Three bait attractants were used: canned sardines, fresh meat (beef mince), and general Elliot trap mix (rolled oats, peanut butter and honey mixed together). Only a single bait attractant was used at each camera. Baits were replenished on a daily basis.

Rosenberg's Goanna were detected on 20 occasions during the five day survey (15 camera days;  $\mu = 1$  animal/0.25 camera day - see Figure 1). However, when considering each of the bait attractants, canned sardines as a bait provided the greatest detectability (15 observations;  $\mu = 1$  animal/0.34 camera day), followed by fresh meat (4 observations;  $\mu = 1$  animal/1.25 camera days) and general Elliot trap mix (1 observation;  $\mu = 1$  animal/5 camera days).

In all canned sardine observations, an individual Rosenberg's Goanna consumed the bait in its entirety. Conversely, fresh meat was only partially consumed, while the general Elliot trap mix was visited by a single individual but not consumed. There appears to be a clear preference for the canned sardines as a bait attractant for Rosenberg's Goanna.

#### ***Trapping***

Cage traps have long been promoted as a successful means of detecting Rosenberg's Goanna. While some researchers have detected Rosenberg's Goanna using cage traps, the authors have had little success despite conducting hundreds of surveys over the past 20 years. With many researchers promoting the use of funnel traps as a successful method of trapping reptiles including goannas (CSC 2008; Denny 2005; Sass 2009; Sass *et al.* in prep.; Thompson and Thompson 2007), the authors established a series of funnel traps with drift fence combinations (20cm tall PVC builders damp course) at ten locations in known habitat in the Liverpool Military Area. Traps were activated over a period of five days in Spring and Autumn over two consecutive years (1200 trap nights). Despite this extensive trapping effort over a two year period, only a single Rosenberg's Goanna was trapped ( $\mu = 1$  animal/1200 trap nights).

## Conclusion

Difficulty in determining the presence of any species of reptile in any landscape type is often compounded by a number of factors (Garden *et al.* 2007; Michael *et al.* 2004). Many species of reptile are cryptic by nature, or can occur at such low densities that they are difficult to detect with even the most exhaustive surveys encountering individuals by chance alone. For example, during reptile surveys in Western Australia, a previously undetected species of reptile was recorded after 25,000 trap nights had already been undertaken (Thompson *et al.* 2003). This is likely the case for the Rosenberg's Goanna given the results of our work.

However, the use of motion-activated cameras to detect the presence of Rosenberg's Goanna with a canned sardine attractant was clearly the most successful method of determining site occupancy ( $\mu = 1$  animal/0.34 camera day), with an additional advantage of cost-efficiency given the obvious savings in vehicle and traditional survey costs.

## Acknowledgments

The data used to make this comparison was obtained from a number of surveys conducted by the authors. We wish to thank Rob Kolano, Department of Defence for site access and logistics at the Liverpool Military Area and Shoalhaven City Council particularly Sandie Jones and Kelie Lowe for project assistance, as well as previous employers of the authors including URS Australia. Portions of this study received financial assistance of the Natural Heritage Trust administered through the Southern Rivers Catchment Management Authority.

All surveys were conducted under the authority of a NSW Office of Environment and Heritage Scientific licence and a Department of Primary Industries Animal Care and Ethics Authority.

## References

- Claridge A. W., Mifsud G., Dawson J. & Saxon M. J. (2004) Use of infrared digital cameras to investigate aspects of the social behaviour of cryptic species. *Wildlife Research* **31**, 645-50.
- Clemann N., Long K., Skurrie D. & Dzuris J. (2005) A trapping survey of small, ground dwelling vertebrates in the Little Desert National Park, Victoria. *Australian Zoologist* **33**, 119-26.
- Cogger H. (2000). *Reptiles and Amphibians of Australia*. Reed New Holland, Sydney.
- CSC. (2008) Reptile Survey of the Proposed Regional Park, Australian Defence Industries Site, St Marys. *A report prepared by Cygnet Surveys and Consultancy for the Department of Environment and Climate Change*.
- DECC. (2007) Threatened and Pest Animals of Greater Sydney. *NSW Department of Environment & Climate Change, Sydney*.
- Denny M. (2005) Reptile Funnel Traps - A road test. *Ecological Consultants Association of NSW Newsletter*.
- Garden J. G., McAlpine C. A., Possingham H. P. & Jones D. N. (2007) Using multiple survey methods to detect terrestrial reptiles and mammals: what are the most successful and cost-efficient combinations? *Wildlife Research* **34**, 218-27.
- Green B., Dryden G. & Dryden K. (1991) Field energetics of a large carnivorous lizard, *Varanus rosenbergi*. *Oecologia* **88**, 547-51.
- King D. & Green B. (1999) *Goannas - the biology of varanid lizards, 2nd edition*. University of New South Wales Press, Sydney.
- Michael D. R., Lunt I. D. & Robinson W. A. (2004) Enhancing fauna habitat in grazed native grasslands and woodlands : use of artificially placed log refuges by fauna. *Wildlife Research* **31**, 65-71.

OEH. (2014a) BioNET: The website for the Atlas of NSW Wildlife: A whole-of-government system for flora and fauna sightings information.

OEH. (2014b) Threatened species, populations and ecological communities of NSW. *NSW Office of Environment & Heritage*.

[www.threatenedspecies.environment.nsw.gov.au](http://www.threatenedspecies.environment.nsw.gov.au).

Rismiller P. D. & McKelvey M. W. (2000) Spontaneous arousal in reptiles? Body temperature ecology of Rosenberg's goanna, *Varanus rosenbergi*. *Life in Cold Blood: Eleventh Internal Hibernation Symposium*, 57-64.

Sass S. (2008) Increasing ecological knowledge and community awareness of the threatened Rosenberg's goanna in the Shoalhaven. *A report prepared by Steve Sass to the Southern Rivers Catchment Management Authority*. <http://www.southern.cma.nsw.gov.au/documents/Rosenbergs%20Goanna%20Report.pdf>.

Sass S., Swan G., Sullivan C. & Parsell S. (in prep.) Whats in a trap? An evaluation of various detection methods for small terrestrial vertebrate fauna in the Cobar Peneplain Bioregion and implications for future fauna surveys and Environmental Impact Assessments. *Australian Zoologist*.

Sass S. (2009) The effectiveness of funnel traps to conduct reptile surveys in the chenopod shrublands of western New South Wales. *Conserving Ecology* **22**, 18-20.

Swan G., Shea G. & Sadlier R. (2004) *Field guide to the reptiles of New South Wales*. Reed New Holland, Sydney.

Swan M. & Watharow S. (2005) *Snakes, Lizards and Frogs of the Victorian Mallee*. CSIRO Publishing, Collingwood.

Thompson G. G. & Thompson S. A. (2007) Usefulness of funnel traps in catching small reptiles and mammals, with comments on the effectiveness of alternatives. *Wildlife Research* **34**, 491-7.

Thompson G. G., Withers P. C., Pianka E. R. & Thompson S. A. (2003) Assessing biodiversity with species accumulation curves; inventories of small reptiles by pit-trapping in Western Australia. *Austral Ecology* **28**, 361-83.

Vincent M. & Wilson S. (1999) *Australian Goannas*. New Holland Publishers, Sydney.

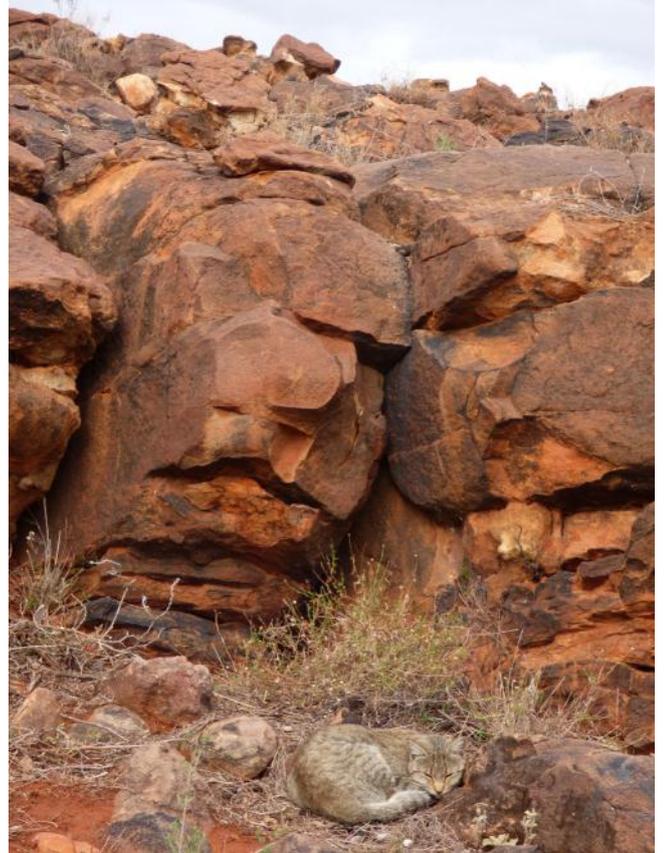
Wilson S. & Swan G. (2013) *A Complete Guide to Reptiles of Australia: Fourth Edition*. Reed New Holland, Sydney.

**Figure 1: Rosenberg's Goanna attracted to canned sardines and detected by a motion-activated infrared camera in northern Sydney.**



# ECA Photo Gallery

## Photo Competition Entries



Barrier Ranges Dragon *Ctenophorus mirrityana*. **Above, and Below Centre :** male. **Below and Bottom Left:** female. Right: A feral cat guarding Barrier Ranges Dragon habitat. (Photos courtesy of Phil Cameron and NSW OEH Western Region)



**Below:** *Diuris Tricolor* at Ulan (Photo courtesy of Phil Cameron).



# ECA Photo Gallery

## Photo Competition Entries



**Above::** Eastern Yellow Robin at Dubbo (*Ady Watson*)

**Below:** Pink-tailed Worm Lizard at Toongi (*Photo courtesy of Phil Cameron*)



**Right:** Scorpion at Toongi (*Photo courtesy of Phil Cameron*)



**Below:** Bustard chick in the 'middle of nowhere' - far western region (*Photo courtesy of Mark Arrow*)

**Below Right:** Speckled Warbler (*Photo courtesy of Ady Watson*)



# FIELD WORK TICKS ME OFF

Stephen J. Ambrose

Director, Ambrose Ecological Services Pty Ltd

## INTRODUCTION

There has been a lot of publicity from within the ECA over the last couple of years about the risks of ticks in Australia transmitting Lyme or Lyme-like diseases and other tick-borne diseases. Consequently, I reviewed my Work Health and Safety & Safety (WH&S) Procedures to take into account this risk alert. In doing so, I realised that I was woefully ignorant about what and how many tick species feed on human blood, their distributions and habitats, life cycles, host species, and the pathogens they carried. Therefore, I headed to the scientific and medical literature to fill in these information gaps to assist me to update my WH&S procedures. I also thought it would be useful to reproduce this information in *Consulting Ecology* for the benefit of other ecological consultants.

## Australian Ticks

There are more than 800 species of ticks worldwide, and they are divided into three families: the Ixodidae (the hard ticks), Argasidae (the soft ticks), and Nuttalliellidae (represented by a single primitive tick species in Africa).

The hard ticks are distinguished from soft ticks by the presence of a scutum (a hard shield). In both the nymph and the adult, a prominent capitulum (head) projects forward from the tick's body, whereas in soft ticks, the capitulum is concealed beneath the body. The external morphologies of hard and soft ticks are shown in Figures 1 and 2, respectively, to assist in the interpretation of species descriptions in the present article.

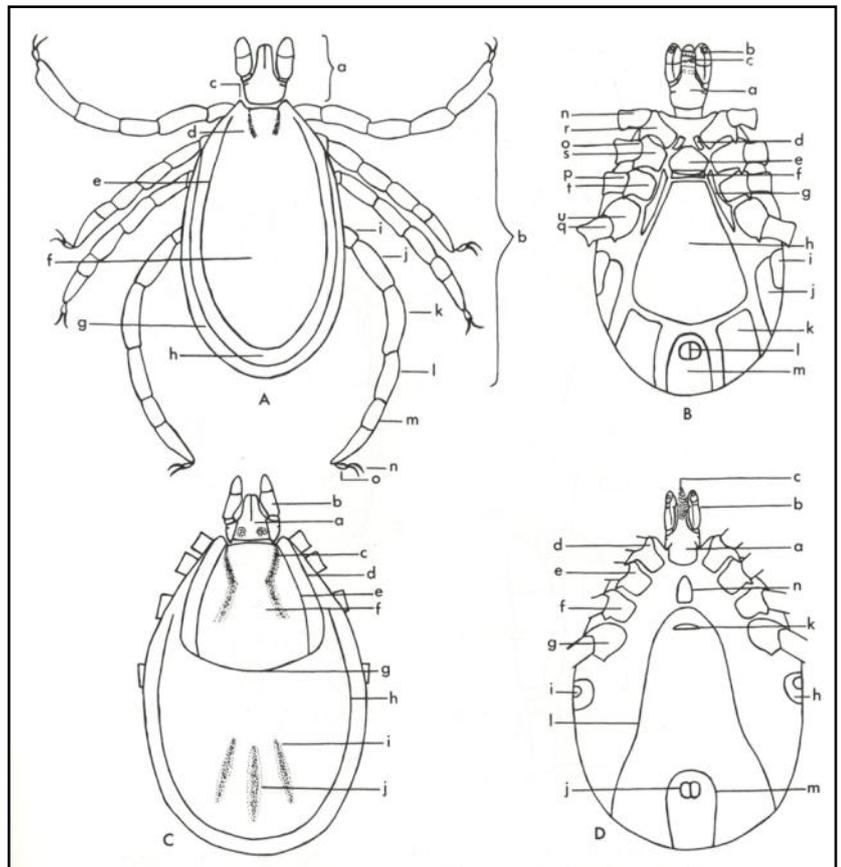
Nuttalliellids are distinguished from hard and soft ticks through a combination of characteristics such as the position of the stigmata (spiracular openings in the integument used for respiration), lack of setae (bristles), and the presence of a strongly corrugated integument and fenestrated plates

Roberts (1970) described in detail the tick species that were known to occur in Australia at the time

of his publication (59 species in total). Seven of these species were soft ticks represented by three genera, *Argas* (three species), *Ornithodoros* (three species) and *Otobius* (one species). The remaining 52 species were hard ticks represented across five genera: *Ixodes* (22 species), *Haemaphysalis* (eight species), *Rhipicephalus* (one species), *Boophilus* (one species), *Amblyomma* (12 species) and *Aponomma* (eight species).

However, Irwin (2012) states that "Australia is home to nearly one hundred known tick species". Taxonomic sources (e.g. Guglielmo *et al* 2010, 2014) have lumped some *Boophilus* species with *Rhipicephalus*, and split *Aponomma* to form a new genus, *Bothriocroton*.

Figure 1 External Morphology of Hard Ticks (from Roberts 1970)



A: *Ixodes* male (dorsal view). a: capitulum; b: body; c: scapula; d: cervical groove; e: lateral groove; f: scutum; g: marginal body fold; h: marginal scutal fold; i: trochanter; j: femur; k: tibia; l: metatarsus; m: tarsus; n: claws; o: pulvillus.

B: *Ixodes* male (ventral view). a: basis capituli; b: palp; c: hypostome; d: jugular plate; e: pregenital plate; f: genital aperture; g: additional accessory plate; h: median plate; i: spiracular plate; j: epimeral plate; k: adanal plate; l: anus; m: anal plate; n: trochanter I (spurred); o: trochanter II (spurred); p: trochanter III (spurred); q: trochanter IV (spurred); r: coxa I (internal and external spur); s: coxa II; t: coxa III; u: coxa IV.

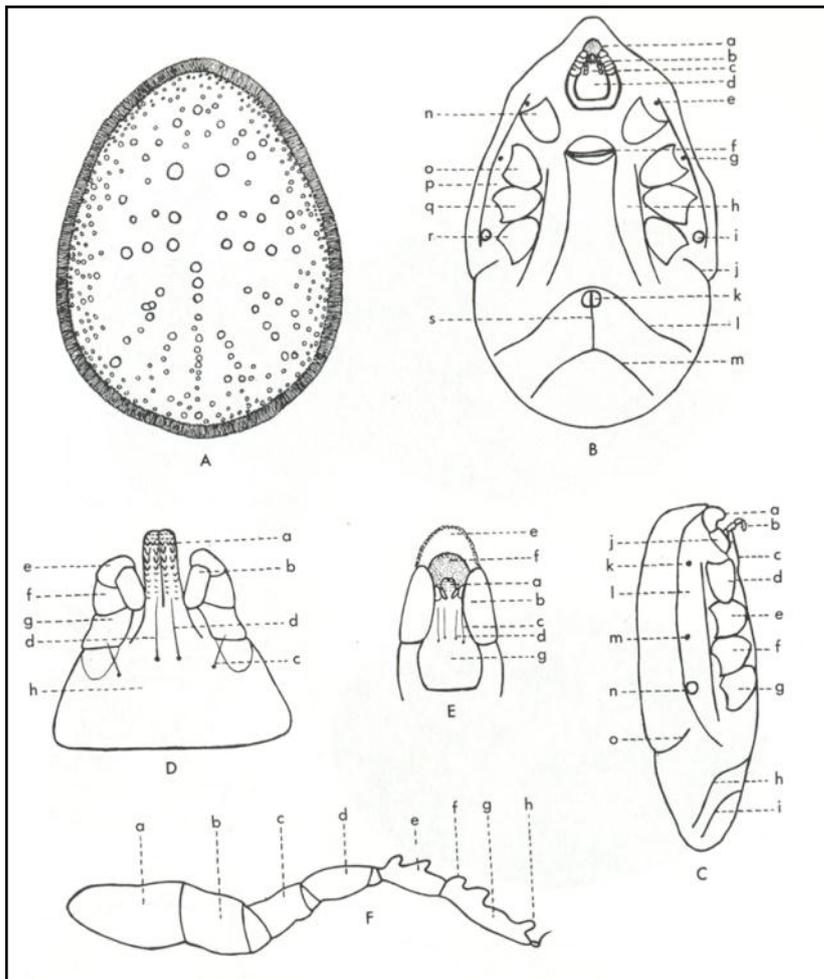
C: *Ixodes* female (dorsal view). a: capitulum; b: palp; c: cervical groove; d: anterolateral margin of scutum; e: lateral carina; f: scutum; g: posterior angle of scutum; h: marginal groove; i: posterolateral groove; j: median groove.

D: *Ixodes* female (ventral view). a: capitulum; b: palp; c: hypostome; d: coxa I; e: coxa II; f: coxa III; g: coxa IV; h: spiracular plate; i: macula; j: anus; k: genital aperture; l: genital groove; m: anal groove; n: sterna plate

Therefore, information about the precise number of hard tick species now occurring in Australia is confusing. For instance, Barker & Walker (2014) state that 28 *Ixodes* species, 18 *Amblyomma* species and 6 *Bothriocroton* species occur in Australia, but did not indicate the number of *Haemaphysalis*, *Rhipicephalus* or *Aponomma* species that occur here.

Barker & Walker (2014) have renamed Australian *Boophilus* as *Bothriocroton* and maintaining it as a separate genus to *Rhipicephalus*.

**Figure 2. External Morphology of Soft Ticks (from Roberts 1970)**



**A:** Dorsal view of *Argas* sp. showing differentiation of body margin and radial arrangement of discs.

**B:** Ventral view of *Ornithodoros* sp. a: hood; b: cheek; c: base of hypostome; d: basis capituli; e: eye; f: genital aperture; g: eye; h: coxal fold; i: spiracle; j: dorsoventral groove; k: anus; l: preanal groove; m: transverse postanal groove; n: coxa I; o: coxa II; p: supracoxal fold; q: coxa III; r: coxa IV; s: median postanal groove.

**C:** Lateral view of *Ornithodoros* sp. a: hood; b: palp; c: basis capituli; d: coxa I; e: coxa II; f: coxa III; g: coxa IV; h: preanal groove; i: transverse postanal groove; j: cheek; k: eye; l: supracoxal fold; m: eye; n: spiracle; o: dorsoventral groove.

**D:** Capitulum of *Argas* sp. a: hypostome; b: palpal article 4; c: postpalpal bristle; d: posthypostomal bristle; e: palpal article 3; f: palpal article 2; g: palpal article 1; h: basis capituli.

**E:** Capitulum of *Ornithodoros* sp. a: hypostome; b: cheek; c: postpalpal bristle; d: posthypostomal bristle; e: hood; f: camerostome; g: basis capituli.

**F:** Leg: a: coxa; b: trochanter; c: femur; d: tibia; e: metatarsus; f: dorsal hump; g: tarsus; h: subapical dorsal protuberance.

There are at least 10 species of hard ticks in Australia that bite humans. These are:

#### *Native Species*

- ◆ Australian Paralysis Tick *Ixodes holocyclus* (Neumann 1899);
- ◆ Southern Paralysis Tick *Ixodes cornuatus* (Roberts 1960);
- ◆ Common Marsupial Tick *Ixodes tasmani* (Roberts 1960);
- ◆ Ornate Kangaroo Tick *Amblyomma triguttatum* (Koch 1844);
- ◆ Wallaby Tick *Haemaphysalis bancrofti* (Nuttall & Warburton 1915);
- ◆ Bandicoot Tick *Haemaphysalis humerosa* (Warburton & Nuttall 1909);
- ◆ Southern Reptile Tick *Bothriocroton (Aponomma) hydrosauri* (Denny 1843).

#### *Introduced Species*

- ◆ Cattle Tick *Rhipicephalus (Boophilus) microplus* (Canestrini 1888);
- ◆ Brown Dog Tick *Rhipicephalus sanguineus* (Latreille 1806); and
- ◆ Bush Tick *Haemaphysalis longicornis (goral)* (Neumann 1901). Note that some sources of information also identify *H. bispinosa* as a tick that bites humans, but this species name is a synonym of *H. longicornis* [http://zipcodezoo.com/Animals/H/Haemaphysalis\\_longicornis/](http://zipcodezoo.com/Animals/H/Haemaphysalis_longicornis/)

Descriptions of the adult, nymph and larval stages, and information about the life-cycle, distribution, habitats, host species, seasonality of abundance, and the diseases carried are presented in Table 1 (Australian Paralysis Tick, Southern Paralysis Tick, Common Marsupial Tick, Ornate Kangaroo Tick, Cattle Tick, Brown Dog Tick, Bandicoot Tick Bush Tick, Wallaby Tick and Southern Reptile Tick. I have reproduced Roberts (1970) descriptions of the life stages of each species in full in these tables because his book remains the definitive work on Australian ticks, the descriptions are essential for accurately identifying tick species while in the field, but the book is now out of print

and difficult to obtain.

At least two Australian soft tick species, the Seabird Soft Tick *Ornithodoros capensis* (Neumann 1901) and Kangaroo Tick *Ornithodoros gurneyi* (Warburton 1926) bites humans. Information about these species is presented in Table 1.

The tables also include images of some of the featured tick species. However, there are some excellent colour plates and line drawings of many of these species in Barker & Walker (2014), which I suggest you use with the information presented in the tables.

Barker & Walker (2014) cautioned that other tick species that infest domesticated animals in Australia have the potential of biting humans, but as yet there are no records of this happening. These species are the Poultry Tick (*Argas persicus*), Robert's Bird Tick (*Argas robertsi*), Spinose Ear Tick (*Otobius megnini*), Wombat Tick (*Bothriocroton auruginans*) and Hirst's Marsupial Tick (*Ixodes hirsti*). None of these species is dealt with further in the present article.

## GENERAL TICK ECOLOGY

Houseman (2013) indicates that soft ticks are more resistant to desiccation than hard ticks, so are most abundant in drier climates and habitats and microhabitats. Conversely, hard ticks are more common in regions, habitats and microhabitats where humidity is relatively high.

While on the ground, hard ticks must locate a humid, cool environment to avoid desiccation. They typically shelter in a moist layer of soil and leaf litter to enter diapause. In contrast, soft ticks have a leathery exoskeleton which is more effective in preventing body water loss, so tend to be nest parasites that feed repeatedly on the same animal or same family group of animals within the nest.

Soil temperature and the number of daylight hours regulate the timing of diapause in hard ticks. At the end of diapauses, the tick emerges from the soil and leaf litter and climbs vegetation to a certain height, based on the stage of development, and waits for a passing host. Adults climb higher and so are more likely to be found on large hosts, whereas larvae and nymphs, which are more susceptible to desiccation and stay closer to the soil, are more likely to come in contact with smaller hosts.

Once on the host, ticks instinctively move upward to a protected site, attach and then feed. Adult soft ticks mate while off the host, but adult hard ticks mate on the host. Males mate with females while the females are feeding. Adult males need to feed on blood for their sperm to mature, but do not need to engorge as much as females and immature stages. After engorgement and mating, the female drops to the ground to lay a large batch of eggs and then dies.

## TRANSMISSION OF DISEASES BY TICKS

Ticks are associated with the transmission of diseases caused by protozoa, spirochaetes, rickettsiae and viruses in all classes of vertebrates, including humans and domesticated animals (Fraser 1970).

Ticks are responsible for the spread of babesiosis (*Babesia argentina*, *Babesia bigemina*), anaplasmosis (*Anaplasma marginale*), spirochaetosis (*Borrelia theileri*) and theileriosis (*Theileria mutans*) in cattle; biliary fever (*Babesia canis*) in dogs; and spirochaetosis (*Borrelia gallinarum*) in poultry.

Many Australian vertebrates that are infested by ticks also act as host reservoirs for tick-borne pathogens that could potentially be passed onto humans. Species groups that are shared tick host species with humans in Australia are listed in Tables 1 to 6.

Confirmed tick-borne diseases of humans occurring in Australia include Q Fever (*Coxiella burnetii*), Queensland Tick Typhus (*Rickettsia australis*) and Flinders Island Spotted Fever (*Rickettsia honei*).

Senanayake *et al* (2012) documented the first recorded case of human babesiosis in Australia (*Babesia microti*) in a patient who died in Canberra in April 2011.

Moreover, some human patients in Australia who exhibit Lyme-like symptoms have tested positive for locally-endemic *Babesia microti* and *Babesia duncani* and/or *Bartonella henselae* in coexistence with *Borrelia* spp. (Mayne 2011).

While there is no proof that the infections documented by Mayne (2011) and Senanayake *et al* (2012) were caused by tick bites, that possibility exists because of the coexistence of humans in areas where other vertebrates are reservoirs for pathogens and tick species are vectors.

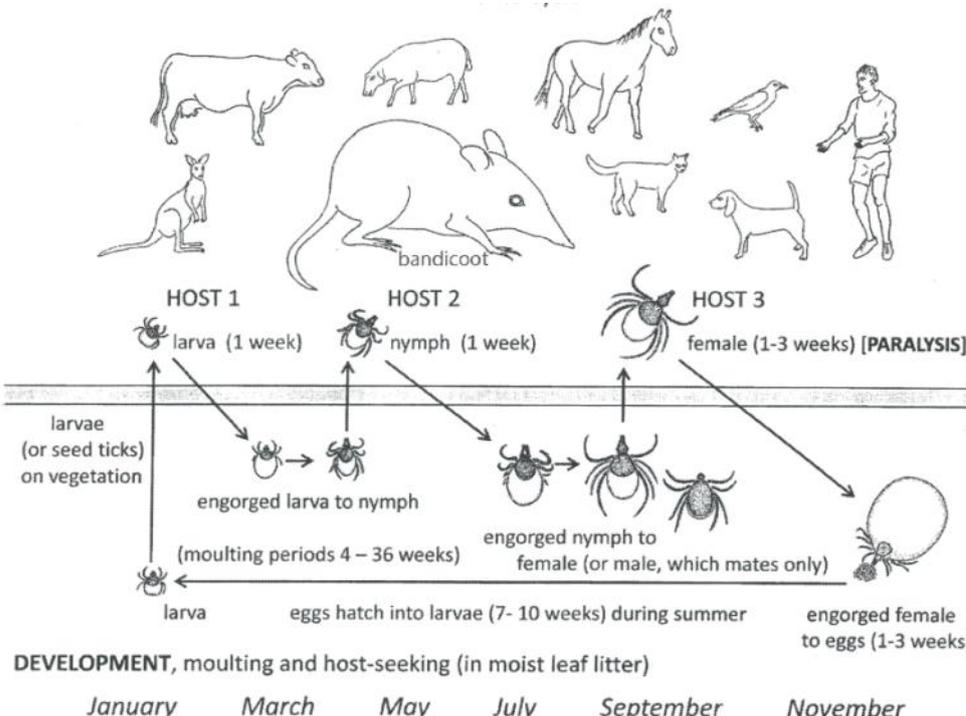
It is also important to note that infected animals can also pass pathogens onto humans without ticks. For instance, Fraser (1970) states that pathogens in the excretions and secretions of infected animals can be transferred to humans, especially if humans come in direct contact with these animals. Humans can also become infected when handling dried animal excrement and body fragments of infected ticks.

## Reducing the Risk of Tick Bites

Measures for reducing the risk of tick bites have been discussed in previous issues of Consulting Ecology. However, it is worth revisiting these procedures in the context of the present article. I hope your WH&S procedures are now compliant with the most recent information about Australian tick-borne pathogens.

1. **Know where to expect ticks:** Hard ticks live in moist and humid environments, particularly in or near wooded or grassy areas; soft ticks live in and around nests, animal burrows and animal scrapes. You may come into contact with ticks during outdoor activities around your home or when walking through vegetation such as leaf litter or shrubs. To avoid ticks, walk in the centre of trails and avoid tall vegetation.
2. **Use a repellent with DEET (on skin or clothing) or permethrin (on clothing and gear):** Repellents containing 20% or more DEET (N, N-diethyl-m-toluamide) can be applied to the skin, and they can protect up to several hours. Always follow product instructions. Parents should apply repellents to their children, taking care to avoid application to hands, eyes, and mouth. Products containing permethrin can be used to treat boots, clothing, and camping gear. Treated items can remain protective through several washings.
3. **Perform daily tick checks:** Check your body for ticks after being outdoors, even in your own yard. Conduct a body check upon return from potentially tick-infested areas by searching your entire body for ticks. Use a hand-held or full-length mirror to view all parts of your body and remove any tick you find. Take special care to check these parts of your body and your child's body for ticks:
  - Under the arms
  - In and around the ears
  - Inside the belly button
  - Back of the knees
  - In and around all head and body hair
  - Between the legs
4. **Check your clothing and pets for ticks** because ticks may be carried into the house on clothing and pets. Both should be examined carefully, and any ticks that are found should be removed. Placing clothes into a dryer on high heat effectively kills ticks.
5. **Remove attached ticks quickly and correctly:** Remove an attached tick using fine-tipped tweezers as soon as you notice it. If a tick is attached to your skin for less than 24 hours, your chance of getting Lyme Disease is extremely small; however, other diseases may be transmitted more quickly.
6. Over the next few weeks, **watch for signs or symptoms of Lyme disease** such as rash, aches, or fever. See a healthcare provider if these develop.
7. **Be alert for fever or rash:** Even if you don't remember being bitten by a tick, an unexpected summer fever or odd rash may be the first signs of a tick-borne disease, particularly if you've been in tick habitat. See your health care provider if these symptoms develop.
8. **Prevent ticks on animals:** Prevent family pets from bringing ticks into the home by limiting their access to tick-infested areas and by using veterinarian-prescribed tick collars or spot-on treatment.
9. **Create tick-safe zones in your yard:** Modify your landscaping to create "Tick-Safe Zones." It's pretty simple. Keep patios, play areas, and playground equipment away from shrubs, bushes, and other vegetation. Regularly remove leaf litter, clear tall grasses and brush around your home, and place wood chips or gravel between lawns and wooded areas to keep ticks away from recreational areas (and away from you). You can also do the following:
  - *Use a chemical control agent:* Effective tick control chemicals are available for use by the homeowner, or they can be applied by a professional pest control expert.
  - *Discourage marsupials, rodents and bovids:* Marsupials, rodents and bovids are significant food sources for adult ticks. Where possible, keep these species away from your home by removing plants and other food sources that attract them. Also constructing physical barriers that may discourage animals from entering your yard and bringing ticks with them.

**Table 1** Summary Information about the Australian Paralysis Tick (*Ixodes holocyclus*), Southern Paralysis Tick (*Ixodes cornuatus*), Common Marsupial Tick (*Ixodes tasmani*) and Ornate Kangaroo Tick (*Amblyomma triguttatum*), Cattle Tick (*Rhipicephalus microplus*) and Brown Dog Tick (*Rhipicephalus sanguinensis*), Bandicoot Tick (*Haemaphysalis humerosa*) and Bush Tick (*Haemaphysalis longicornis*), Wallaby Tick (*Haemaphysalis bancrofti*) and Southern Reptile Tick (*Bothriocroton hydrosauri*), Seabird Soft Tick (*Ornithodoros capensis*) and Kangaroo Soft Tick (*Ornithodoros gurneyi*)

	<b>Australian Paralysis Tick <i>Ixodes holocyclus</i> Neumann, 1899</b>
<p>Known Stages</p>	<p>Male, female, nymph, larva. Each stage is dependent on a host to complete its development. From Barker &amp; Walker (2014):</p>  <p style="text-align: center;"><b>DEVELOPMENT, mouling and host-seeking (in moist leaf litter)</b></p> <p style="text-align: center;">January      March      May      July      September      November</p>
<p>Zoogeographic region</p>	<p>Australasian</p>
<p>Description of Adult Male (extract from Roberts 1970)</p> 	<p>Body measurements less than 3.0 x 2.5 mm; lateral grooves completely encircling scutum, no lateral carinae; punctuations fine; basis capituli punctate dorsally, palpi short and very broad; hypostome dentition 2/2, with rounded teeth; anal plate bluntly pointed behind; adanal plate curving inwardly to a point; coxae with well-defined spurs decreasing in size posteriorly; trochanters III and IV frequently with small, ventral spurs. <b>Body:</b> Oval, sometimes broadly so, 1.9 x 1.6 mm- 3.2 x 2.3 mm; marginal body fold narrow but prominent; hairs dorsally sparse medianly, more numerous on marginal body fold. <b>Capitulum:</b> Length 0.51- 0.65 mm in width, surface punctate; posterior margin straight; no cornua; posterolateral margins slightly divergent anteriorly; basis ventrally narrowing to the straight posterior margin, surface with a short anterolateral ridge. Palps short and broad; article 1 rounded and a little salient laterally, ventrally with a transverse rounded flange continuous with ridge on basis; articles 2 and 3 with no apparent suture, 0.33- 0.40 mm in length, almost twice as long as broad, rounded distally, hairs moderate in number, some long hairs ventrally. Hypostome short and broad, 0.25- 0.28 mm in length, narrowing and shallowly rounded distally; dentition 2/2 of large rounded teeth, some small teeth distally and crenulations basally. <b>Scutum:</b> Oval, convex, only a little smaller than body. Lateral grooves deep and completely encircling the scutum, anteriorly somewhat linear and may simulate mild lateral carinae. Punctuations fine, usually most numerous submarginally and anteromedianly; pseudo-scutum sometimes faintly apparent. Cervical grooves, short, shallow. Emargination moderate. Scapulae blunt. <b>Genital aperture:</b> On a level with anterior margin of coxa III, sometimes at level of 2nd inter-coxal space. <b>Ventral plates:</b> Pregenital plate wider than long; median plate 1.5 x 1.2 mm, the width posteriorly about 3/4 of the length; anal plate 0.75 x 0.50 mm, anterior margin straight or mildly curved, pointed posteriorly; adanal plates curving to points near the point of the anal plate; plates with scattered punctuations and hairs. <b>Spiracular plate:</b> Elongate, oval, narrow posteriorly, the longer axis directed anteriorly, about 0.50- 0.53 mm in length. <b>Legs:</b> Length moderate. Coxae practically contiguous, with a row of long hairs near posterior margin; posterointernal angles of coxae I and II may be somewhat sharp but not salient; all coxae with an external spur, strongest and bluntly pointed on coxa I, smallest on coxa IV. Trochanters III and IV with a small, dark ventral spur, only a tuberosity on II. Tarsi ending somewhat abruptly; length of tarsus I 0.65- 0.71 mm, and of tarsus IV 0.62- 0.70 mm.</p>

**Australian Paralysis Tick**  
***Ixodes holocyclus* Neumann, 1899**

Description of Adult Female (extract from Roberts 1970)



A very large tick when fully engorged; scutum about as long as broad and broadest a little posterior to mid length, with strong lateral carinae; capitulum relatively long porose areas deep, cornua usually absent, but when present at most only mild and rounded; auriculae present; hypostome lanceolate, dentition mainly 3/3; no sternal plate; anal grooves meeting at a point behind; all coxae with an external spur decreasing in size posteriorly; trochanters III and IV usually with small, pointed ventral spurs. **Body:** Unfed specimens, oval, flat, yellowish, 2.6 x 1.1 mm - 3.8 x 2.6 mm; marginal groove well developed and continuous; hairs small, scattered, most numerous in region of marginal fold. Semi-engorged specimens frequently with body widest behind coxa IV and with a waist at level of spiracles. Fully engorged specimens broadly oval, attaining 13.2 x 10.2 mm, living ticks with blue-grey alloscutum, the dorsum light in colour, a dark band in region of marginal groove. **Capitulum:** Length 1.00-1.035 mm. Basis dorsally 0.60- 0.68 mm in width, the lateral submarginal fields swollen and frequently delimited from the depressed, median field by ill-defined carinae; posterior margin sinuous, posterolateral angles swollen, sometimes mildly salient; porose areas large, deep subcircular or oval, the longer axis directed anteriorly, interval frequently depressed, at most about the width of one; basis ventrally with posterior margin rounded and with well-defined, blunt, retrograde auriculae. Palps long and slender, some long hairs ventrally; article I rounded and somewhat salient laterally, inner 'ring' with dorsal tongue-like prolongation and ventrally semicircular and plate-like, the posterior margin of the plate extending beyond the palp; articles 2 and 3 with no apparent suture, 0.75- 0.85 mm in length and about four times as long as wide, narrowly rounded distally. Hypostome lanceolate and bluntly pointed; dentition mainly 3/3, the innermost file of small, spaced teeth, basally 2/2. **Scutum:** As wide as or a little wider than long, widest a little posterior to mid length, 1.6 x 1.7 mm- 2.4 x 2.4 mm, flat medianly, convex external to the long, strong lateral carinae; anterolateral margins practically straight, posterolateral margins mildly concave; posterior angle broadly rounded. Punctations numerous, fine, sometimes a little coarser medianly and laterally, shallow rugae frequently present posteriorly. Cervical grooves well defined but short. Emargination moderate. Scapulae blunt. **Genital aperture:** On a level with coxa IV, but in engorged specimens sometimes just posterior to this level. **Anal grooves:** Rounded anteriorly, curving behind anus to meet in a somewhat elongate point. **Spiracular plate:** Subcircular, greatest dimension 0.40- 0.45 mm. **Legs:** Coxae smooth, I and II sometimes with mild rounded ridges externally, each with a row of long hairs posteriorly and an external spur, longer and more pointed than in male, and decreasing in size posteriorly. Trochanter IV (and sometimes III) frequently with a small, ventral spur. Tarsi tapering a little abruptly; length of tarsus I 0.70- 0.80 mm, and of tarsus IV 0.60- 0.78 mm.

Description of Nymph (extract from Roberts 1970)



Capitulum as in female, hypostome dentition mainly 2/2, 3/3 distally; scutum about as long as wide with lateral carinae; sternal plate present, oval; anal grooves converging posteriorly but remaining narrowly open; legs as in female. **Body:** Oval with fine parallel striae and some scattered pale hairs; 1.2 x 0.85 mm (unfed) to 3.5 x 2.5 mm (engorged); marginal grooves well developed and complete in unfed specimens. **Capitulum:** Length 0.40- 0.43 mm. Basis dorsally 0.23- 0.25 mm in width; posterior margin straight; posterolateral angles not salient; auriculae well defined. Palps as in female; articles 2 and 3 0.30- 0.32 mm in length. Hypostome lanceolate and bluntly pointed; dentition 2/2, 3/3 distally. **Scutum:** About as wide as long, 0.61 x 0.63 mm - 0.71 x 0.70 mm. Lateral carinae well developed. Punctations few, shallow, scattered. Cervical grooves apparent, continuing to mid-scutal region as superficial depressions. **Sternal plate:** Oval, 0.27- 0.30 mm in length and a little more than twice as long as wide. **Spiracular plate:** Subcircular, greatest diameter about 0.14 mm. **Legs:** Coxae armed as in female. Tarsus I tapering gradually, other tarsi more abruptly; length of tarsi I and IV about 0.28 mm.

Description of Larva (extract from Roberts 1970)

Capitulum with slender palpi, hypostome rounded apically, dentition 2/2; scutum about as long as wide, with faint lateral carinae; all coxae with small, external spurs. **Body:** Broadly oval, 0.5 x 0.4 mm (unfed) to 1.15 x 1.0 mm (engorged) **Capitulum:** About 0.2 mm in length, basis triangular, about 0.16 mm wide, palpi elongate and slender. Hypostome apically rounded, 0.14 mm in length, dentition 2/2 of 10-12 teeth, the teeth of the inner file blunt and small, some minute denticles apically. **Scutum:** About as long as broad, 0.31 by 0.32 mm and widest a little anterior to mid-length, lateral carinae present but faint; anterolateral margins usually convex and posterolateral margins concave; cervical grooves short but well defined. **Anal grooves:** Ill defined anteriorly and do not converge behind **Legs:** Coxae with small external spurs; tarsus I 0.14 mm in length, tarsus IV 0.14 mm in length

Description of Egg (extract from Roberts 1970)



Adult females lay large numbers of eggs (between 2000 and 6000) in leaf and branch litter, under the scaly or fibrous bark of certain trees and shrubs, or in dense fine foliage near the tips of branches. They utilise a wax-like substance to make a cohesive mass of eggs and attach these at the selected site. A small fraction of the eggs survive and hatch to larvae after 40–110 days incubation. Development occurs with suitable warmth and high humidity (e.g. moist leaf litter).

<b>Australian Paralysis Tick</b> <i>Ixodes holocyclus</i> Neumann, 1899	
Ecoregions	Temperate broadleaf and mixed forests of eastern Australia. Known from Normanton, north Qld to the Lakes Entrance, eastern Vic. In NSW, it rarely ranges more than 16 km from the coast, but there are some records from around Armidale. It is abundant within the Sydney area. Nymphs have been found on birds in the ACT, but may have been transported there by long-range flights of their hosts.
Natural Hosts	<p><b>MAMMALS</b></p> <p><b>Artiodactyla:</b> Bovidae</p> <p><b>Carnivora:</b> Felidae, Canidae (including the Dingo <i>Canis familiaris</i>).</p> <p><b>Dasyuromorphia:</b> Dasyuridae (Spotted-tailed Quoll <i>Dasyurus maculatus</i>, Yellow-footed Antechinus <i>Antechinus flavipes</i>, Brush-tailed Phascogale <i>Phascogale tapoatafa</i>), Slender-tailed Dunnart (<i>Sminthopsis murina</i>).</p> <p><b>Diprotodontia:</b> Macropodidae (Red-legged Pademelon <i>Thylogale stigmatica</i>, Black-striped Wallaby <i>Wallabia dorsalis</i>, Red-necked Wallaby <i>Wallabia rufogrisea</i>, Swamp Wallaby <i>Wallabia bicolor</i>, Lumholtz's Tree-kangaroo <i>Dendrolagus lumholtzi</i>), Phalangeridae (Common Brushtail Possum <i>Trichosurus vulpecula</i>, Mountain Brushtail Possum <i>Trichosurus caninus</i>), Petauridae, Phascolarctidae (Koala <i>Phascolarctos cinereus</i>), Potoroidae (Rufous Bettong <i>Aepyprymnus rufescens</i>), Vombatidae.</p> <p><b>Monotremata:</b> Tachyglossidae (Kangaroo Island Echidna <i>Tachyglossus aculeatus</i>).</p> <p><b>Peramelemorphia:</b> Peramelidae (Southern Brown Bandicoot <i>Isodon obselus</i>, Northern Brown Bandicoot <i>Isodon macrourus</i>, Long-nosed Bandicoot <i>Perameles nasuta</i>)</p> <p><b>Rodentia:</b> Muridae (Black Rat <i>Rattus rattus</i>, Norwegian Rat <i>Rattus norvegicus</i>, Bush Rat <i>Rattus fuscipes</i>, Dusky Field Rat <i>Rattus sordidus</i>, Pale Field Rat <i>Rattus tunneyi</i>, False Water Rat <i>Hydromys chrysogaster</i>, Giant White-tailed Rat <i>Uromys caudimaculatus</i>, Fawn-footed Melomys <i>Melomys cervinipes</i>, Grassland Melomys <i>Melomys lutillus littoralis</i>, House Mouse <i>Mus musculus</i>).</p> <p><b>BIRDS</b></p> <p><b>Cuculiformes:</b> Cuculidae.</p> <p><b>Passeriformes:</b> Acanthizidae, Artamidae (Australian Magpie <i>Gymnorhina tibicen</i>), Corvidae (Australian Raven <i>Corvus coronoides</i>), Cracticidae (Pied Butcherbird <i>Cracticidae nigrogularis</i>), Meliphagidae, Pittidae (<i>Pitta</i> sp.), Psittacidae (Crimson Rosella <i>Platycercus elegans</i>), Ptilinorhynchidae,</p>
Human Infestation	Yes
Human Disease Risks	<p><b>BACTERIAL DISEASES</b></p> <p>Rickettsial Spotted Fever (Queensland Tick Typhus). Pathogen: <i>Rickettsia australis</i>.</p> <p>Flinders Island Spotted Fever. Pathogen: <i>Rickettsia honei</i>.</p> <p>Q-Fever. Pathogen: <i>Coxiella burnetii</i></p> <p>Lyme-like Disease in Australia. Pathogens not yet identified with certainty. <i>Borrelia burgdorferi</i>, usually co-existing with <i>Babesia</i>, <i>Bartonella</i> and/or <i>Ehrlichia</i> spp., is known to cause Lyme Disease in the Northern Hemisphere. Recent research has shown some patients in Australia who exhibit Lyme-like symptoms have tested positive for locally-endemic <i>Babesia microti</i> and <i>Babesia duncani</i> and/or <i>Bartonella henselae</i> in coexistence with <i>Borrelia</i> spp.</p> <p><b>VIRAL DISEASES</b></p> <p>So far, no human viruses have been isolated from <i>Ixodes holocyclus</i>.</p> <p><b>PROTOZOAL DISEASES</b></p> <p>So far, no infectious protozoans have been isolated from <i>Ixodes holocyclus</i>.</p>
Seasonality	Each life stage can be present throughout the year, although adults are more abundant in the spring and the early summer months, larvae in mid to late-summer, and nymphs during winter.
<b>Southern Paralysis Tick</b> <i>Ixodes cornuatus</i> Roberts 1960	
Known Stages	Male, female, nymph, larva. Nothing is known about the life cycle and seasonality of this species. Barker & Walker (2014) suggest that the life cycle may be similar to <i>I. holocyclus</i> .
Zoogeographic region	Australasian.
Description of Adult Male (extract from Roberts 1970)	<b>Body:</b> Oval, 4.3 x 2.8 mm to 4.5 x 2.9 mm; marginal body fold well developed throughout, narrow anteriorly; hairs dorsally sparse, more numerous laterally and ventrally. <b>Capitulum:</b> Length 0.69-0.72 mm. Basis dorsally 0.59-0.64 mm in width, surface mildly punctate; posterior margin straight; no cornua; basis ventrally narrowing to posterior margin and with well defined, transverse ridges extending inwards from palpal insertions. Palps short and stout; article 1 rounded and salient laterally, wider than long, ventrally with a pronounced, subcircular ridge; articles 2 and 3 without apparent suture, 0.44-0.46 mm in length and not quite twice as long as wide, a row of some long hairs ventrally. Hypostome short and broad, rounded distally; dentition 2/2 of four or five large rounded teeth, a few small, rounded teeth distally, some crenulations basally.

	<b>Southern Paralysis Tick</b> <i>Ixodes cornuatus</i> Roberts 1960
Description of Adult Male (extract from Roberts 1970) <i>continued</i>	<b>Scutum:</b> Oval, convex, the lateral grooves continuous, deep except anteriorly, no apparent lateral carinae. Punctations sparse, usually fine, sometimes a little coarser and more numerous laterally and anteriorly. Cervical grooves short, superficial posteriorly. Emargination moderate. Scapulae blunt. <b>Genital Aperture:</b> On a level with coxa III. <b>Ventral Plates:</b> Pregonital plate hexagonal, wider than long; median plate 2.0 x 1.6 to 2.3 x 1.4 mm; anal plate 0.9 x 0.6 mm to 1.0 x 0.65 mm, pointed posteriorly; adanal plate with the external margin almost straight, then curving inwardly posteriorly to a blunt point which reaches the point of the anal plate; punctations fine and scattered; hairs scattered. <b>Spiracular Plate:</b> Large, elongate oval and narrower posteriorly, the longer axis directed anteriorly, greatest dimension 0.70-0.72 mm. <b>Legs:</b> Relatively stout and long, with many short hairs and pale articulations. Coxae almost contiguous and with a row of long hairs near posterior margin; all coxae with an external, bluntly pointed spur, strongest on coxa I, smallest on coxa IV. Trochanters ventrally with an external spur, small on I, rounded on II, most pronounced and pointed on III and IV. Tarsi tapering abruptly, with small, but distinct subterminal humps most prominent on tarsi II-IV; length of tarsus I 0.80-0.82 mm, and of tarsus IV 0.79 mm.
Description of Adult Female (extract from Roberts 1970)	<b>Body:</b> Unfed specimens oval; 3.5 x 2.5 mm to 4.7 x 3.3 mm; marginal grooves well developed and complete; hairs dorsally, small, sparse, and mainly lateral, more numerous ventrally; engorged specimens attaining 12.7 x 9.0 mm. <b>Capitulum:</b> Length 1.1-1.5 mm. Basis dorsally 0.80-0.85 mm in width, swollen laterally and frequently also at base of mouthparts to leave median field depressed; posterior margin strongly sinuous, concave medianly, with well developed, bluntly pointed cornua; porose areas large, deep, oval, internal narrow; basis ventrally with strong, blunt, somewhat retrograde auriculae and a curved posterior margin. Palps long and slender; article 1 rounded and salient laterally, the inner "ring" ventrally large, with a subcircular plate and dorsally with a tongu-like process extending for about half the length of the insertion; articles 2 and 3 apparently fused, 1.0-1.2 mm in length and almost four times as long as wide. Hypostome lanceolate and bluntly pointed, about 0.96 mm in length; dentition mainly 3/3, the innermost file of small teeth and somewhat crowded distally, 2/2 basally. <b>Scutum:</b> A little longer than wide and widest at about mid-length, 2.4 x 2.3 mm to 2.7 x 2.6 mm, median field mildly convex and separates from the strongly convex lateral fields by long, strong carinae; anterolateral margins a little sinuous, posterolateral margins a little concave, posterior angle rounded. Punctations fine, not numerous, fairly regularly distributed. Cervical grooves shallow and somewhat superficial posteriorly to reach a little beyond mid-scutal length. Emargination moderate. Scapulae blunt. <b>Genital Aperture:</b> On a level with coxa IV. No sternal plate. <b>Anal Grooves:</b> Rounded anteriorly, curving posteriorly to meet in a point. <b>Spiracular Plate:</b> Subcircular, greatest dimension 0.50-0.55 mm. <b>Legs:</b> Relatively stout, length moderate. Coxae flat, each with a row of hairs near posterior margin and well-defined external spur, the spur on coxa I strong and pointed, that on coxa IV small. Trochanters with rounded spurs, small on I. Tarsi stout, each with a small subterminal hump and terminating abruptly; length of tarsus I 1.0-1.1 mm, and of tarsus IV 0.90-0.97 mm.
Description of Nymph (extract from Roberts 1970)	Not known.
Description of Larva (extract from Roberts 1970)	No description provided.
Description of Egg (extract from Roberts 1970)	No description provided.
Ecoregions	Occurs in the southern coastal areas of NSW, eastern Victoria as far west as Dandenong and Mt Buffalo, and in Tasmania. Nothing is known about the preferred habitat of this species.
Natural Hosts	<b>MAMMALS</b> <b>Artiodactyla:</b> Bovidae <b>Carnivora:</b> Felidae, Canidae (including the Dingo <i>Canis familiaris</i> ). <b>Diprotodontia:</b> Macropodidae (Tasmania Pademelon <i>Thylogale billardierii</i> , Phalangeridae (Common Brushtail Possum <i>Trichosurus vulpecular</i> ), Potoroidae (Tasmania Bettong <i>Bettongia gaimardi cuniculus</i> ), Vombatidae (Common Wombat <i>Vombatus ursinus</i> ). <b>Peramelemorphia:</b> Peramelidae (Southern Brown Bandicoot <i>Isodon obseus</i> , Eastern Barred Bandicoot <i>Perameles gunii</i> ) <b>Rodentia:</b> Muridae (Black Rat <i>Rattus rattus</i> , Swamp Rat <i>Rattus lutreolus</i> , Bush Rat <i>Rattus fuscipes</i> , House Mouse <i>Mus musculus</i> ). <b>BIRDS</b> <b>Passeriformes:</b> Acanthizidae (Tasmanian Scrubwren <i>Sericornis humilis</i> ), Artamidae (Black Currawong <i>Strepera fuliginosa</i> ), Cracticidae (Grey Butcherbird <i>Cracticidae torquatus</i> ), Pachycephalidae (Grey Shrike-thrush <i>Colluricincla harmonica</i> ).

Human Infestation	Yes
Human Disease Risks	Not studied, but Barker & Walker (2014) suggest that <i>I. cornuatus</i> may carry the same or similar pathogens to <i>I. holocyclus</i> .
Seasonality	Not known.
	<b>Common Marsupial Tick</b> <i>Ixodes tasmani</i>
Known Stages	Male, female, nymph, larva.  Little is known of the life history of <i>I. tasmani</i> . Roberts (1970) suggests that it is a three-host tick and that the males appear to be restricted to the host habitat. The larva, nymph and female have been collected from the one host.
Zoogeographic region	Australasian.
Description of Adult Male (extract from Roberts 1970)	<b>Body:</b> Oval, widest about mid-body region, narrowing posteriorly; marginal body fold well developed, with longitudinal furrows, commencing near a level with coxa III, widening laterally and narrowing again posteriorly; hairs pale, short, numerous ventrally, few dorsally. <b>Capitulum:</b> Short, 0.34-0.41 mm in length. Basis dorsally with surface punctate, 0.32-0.40 mm in width; posterior margin straight or almost so; no cornua; posterolateral margins also practically straight, divergent anteriorly; basis ventrally widest just posterior to palpal insertions, the lateral margins concave, the posterior margin shallowly rounded; no auriculae. Palps short, with few hairs; article 1 enlarged, much wider than long, extending inwardly beyond base of article 2, ventrally with a pronounced ridge which is continuous with a similar short ridge on basis; articles 2 and 3 indistinctly separated, short and broad, about 0.3 mm in length, broadening rapidly from base to a width of about half the length, rounded apically. Hypostome 0.19 mm in length, somewhat broad and rounded apically; dentition 4/4 and 3/3 of small teeth distally, then 2/2 of five rows of large rounded teeth and a few rows of shallow, ridge-like teeth basally. <b>Scutum:</b> Elongate oval, 2.3 x 1.4 mm to 3.1 x 1.4 mm, convex, widest in region of coxae II and III, narrowly rounded posteriorly. No lateral grooves or carinae. Punctations numerous, crowded, evenly distributed, mostly moderate in size with some confluency. Cervical grooves distinct for only a short distance anteriorly, then continuing posteriorly as faint shallow depressions. Width between the scapulae narrow. <b>Genital Aperture:</b> On a level with second intercoxal space. <b>Ventral Plates:</b> Pregenital plate subhexagonal, wider than long; medianplate almost twice as long as wide, 1.00 x 0.60 mm to 1.60 x 0.82 mm; anal plate about twice as long as wide, 0.70 x 0.30 to 0.80 x 0.38 mm; anterior margin almost straight, lateral margins practically straight, subparallel; adanal plate almost straight-sided and a little wider anteriorly than posteriorly; all plates except pregenital plate with many punctations and minute hairs. <b>Spiracular Plate:</b> Suboval; the greatest dimension 0.26-0.29 mm, directed anteriorly. <b>Legs:</b> Length moderate, hairs few. Coxae unarmed, all with syncoxae, hairs few. Tarsi tapering somewhat abruptly; length of tarsus I 0.42-0.56 mm, and of tarsus IV 0.46-0.55 mm.
Description of Adult Female (extract from Roberts 1970)	<b>Body:</b> Unfed specimens flat, oval, widest just anterior to spiracular plates, 2.4 x 1.4 mm to 2.6 x 1.6 mm; marginal median and posterolateral grooves well defined. Fully fed specimens attaining 11.8 x 5.6 mm; marginal grooves no longer apparent. <b>Capitulum:</b> Short, 0.48-0.66 mm in length. Basis dorsally 0.38-0.51 mm wide; posterior margin straight or mildly curved; posterolateral angles may be somewhat swollen, but no cornua; posterolateral margins divergent anteriorly; porose areas moderately large, usually oval and convergent anteriorly, the pits relatively large, interval wide and depressed; basis ventrally broadly rounded posteriorly; no auriculae. Palps short with a few hairs; article 1 greatly enlarged, extending inwardly and anteriorly to ensheath basal proportion of mouthparts, dorsally subrectangular, ventrally somewhat subtriangular and strongly salient laterally, the salience frequently visible dorsally, the basal margin extending posteriorly as a bluntly pointed prolongation to cover the anterior portion of coxa I; articles 2 and 3 without any apparent suture, clavate, 0.40-0.45 mm in length, usually convergent distally. Hypostome spatulate, 0.22-0.24 mm in length, broadly rounded distally; dentition with rather crowded teeth, 4/4 distally, then 3/3 and sometimes some rows of 2/2 minute teeth basally, rarely 4/4 throughout. <b>Scutum:</b> Wider than long, 1.2 x 1.3 mm to 1.6 x 1.8 mm, widest a little posterior to mid-length; anterolateral margins straight or mildly curved; posterolateral margins mildly convex, sometimes sinuous, but occasionally concave and almost indented; posterior angle broadly rounded. Punctations numerous, usually denser, coarser and frequently confluent in lateral and cervical fields, sometimes relatively coarse throughout. No lateral carinae. Cervical grooves usually well defined anteriorly, shallow posteriorly and frequently attaining scutal margin. Enargination moderate. Scapulae bluntly pointed. <b>Genital Aperture:</b> On a level with coxa II in unfed specimens, but moving to a level with second intercoxal space as engorgement proceeds. <b>Anal Grooves:</b> Broadly rounded anteriorly, curving a little convergently posteriorly. <b>Spiracular Plate:</b> Moderate in size, oval, the longer axis transverse, 0.35 x 0.42 mm. <b>Legs:</b> Moderate in length, hairs few. Coxae unarmed, flat, all coxae with syncoxae. Tarsi tapering somewhat abruptly; length of tarsus I 0.50-0.57 mm, and of tarsus IV 0.53-0.58 mm.

	<b>Common Marsupial Tick</b> <i>Ixodes tasmani</i>
Description of Nymph (extract from Roberts 1970)	<b>Body:</b> Marginal grooves distinct in unfed specimens, not apparent in fed specimens; 1.7 x 1.0 mm to 2.9 x 2.1 mm: hairs short, sparse. <b>Capitulum:</b> Length about 2.6 mm. Basis dorsally about 0.24 mm wide, as in female. Palps as in female but posterior prolongation of basoventral margin of article I more pronounced and more pointed; articles 2 and 3 about 0.17 mm in length. Hypostome about 0.16 mm in length; dentition 2/2 of about eight teeth. <b>Scutum:</b> Broader than long, 0.48 x 0.70 mm to 0.53 x 0.76 mm. Punctations numerous, coarse, fairly evenly distributed and imparting a somewhat rugose appearance to surface. Cervical grooves distinct, elongate and usually attaining scutal margins. <b>Anal Grooves:</b> Subparallel posteriorly in unfed specimens, curving gently convergently in fully fed specimens. <b>Spiracular Plate:</b> Oval; longer axis transverse, about 0.14 mm. <b>Legs:</b> As in female.
Ecoregions	According to Roberts (1970), <i>I. tasmani</i> is the most common and widespread of all Australian <i>Ixodes</i> . Abundant and widespread in Tas and Vic. In NSW, there are records from throughout the entire coastal and subcoastal areas with inland extensions to Moree, Dubbo and Kosciusko. Known in Qld 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 south-eastern SA and south-west WA.
Natural Hosts	<b>MAMMALS</b> . <b>Carnivora:</b> Canidae (Domestic Dog <i>Canis familiaris</i> ), Felidae (Domestic Cat <i>Felis catus</i> ). <b>Dasyuromorphia:</b> Dasyuridae (Agile Antechinus <i>Antechinus agilis</i> , Yellow-footed Antechinus <i>Antechinus flavipes</i> , Tasmanian Dusky Antechinus <i>Antechinus swainsonii swainsonii</i> , Dusky Antechinus <i>Antechinus swainsonii mimetes</i> , Brush-tailed Phascogale <i>Phascogale tapoatafa</i> , White-footed Dunnart <i>Sminthopsis leucopus</i> , Common Dunnart <i>Sminthopsis murina</i> ). <b>Diprotodontia:</b> Burramyidae (Little Pygmy-possum <i>Cercatetus lepidus</i> ), Macropodidae (Pearson Island Rock-wallaby <i>Petrogale lateralis pearsoni</i> , Tasmanian Pademelon <i>Thylogale billardieri</i> , Red-legged Pademelon <i>Thylogale stigmatica</i> , Red-necked Wallaby ( <i>Macropus rufogriseus</i> ), Petauridae (Leadbeater's Possum <i>Gymnobelideus leadbeateri</i> , Yellow-bellied Glider <i>Petaurus australis</i> , Sugar Glider <i>Petaurus breviceps</i> , Greater Glider <i>Petauroides volans</i> ), Phalangeridae (Short-eared Brushtail Possum <i>Trichosaurus caninus</i> , Common Brushtail Possum <i>Trichosurus vulpecula vulpecula</i> , Tasmanian Brushtail Possum <i>Trichosurus vulpecula fuliginosus</i> ), Phascolarctidae (Koala <i>Phascolarctos cinereus</i> ), Potoroidae (Long-nosed Potoroo <i>Potorous tridactylus</i> , Tasmanian Bettong <i>Bettongia gaimardi</i> ), Pseudocheiridae (Green Ringtail Possum <i>Pseudocheirops archeri</i> , Common Ringtail Possum <i>Pseudocheirops peregrinus</i> ), Vombatidae (Common Wombat <i>Vombatus ursinus</i> ), <b>Lagomorpha:</b> Leporidae (Rabbit <i>Oryzctolagus cuniculus</i> ). <b>Monotremata:</b> Tachyglossidae (Short-beaked Echidna <i>Tachyglossus aculeatus</i> ) <b>Peremelemorphia:</b> Peramelidae (Eastern Barred Bandicoot <i>Perameles gunnii</i> , Long-nosed Bandicoot <i>Perameles nasuta</i> , Southern Brown Bandicoot <i>Isoodon obesulus</i> , Northern Brown Bandicoot <i>Isoodon macrourus</i> ) <b>Perissodactyla:</b> Equidae. <b>Rodentia:</b> Muridae (Long-tailed Mouse <i>Pseudomys higginsii</i> , Broad-toothed Rat <i>Mastacomys fuscus</i> , Fawn-footed Melomys ( <i>Melomys cervinipes</i> , White-tailed Rat <i>Uromys</i> sp., Canefield Rat <i>Rattus sordidus</i> , Bush Rat <i>Rattus fuscipes</i> , Swamp Rat <i>Rattus lutreolus</i> , Brown Rat <i>Rattus norvegicus</i> , Black Rat <i>Rattus rattus</i> , Water Rat <i>Hydromys chrysogaster</i> ).
Human Infestation	Yes.
Human Disease Risks	Tick paralysis in domesticated animals and in wildlife has not been associated with <i>I. tasmani</i> . However, Barker & Walker (2014) indicate that the DNA of five different types of bacteria have been found in <i>I. tasmani</i> : a spotted fever group <i>Rickettsia</i> sp., <i>Rickettsia tasmanensis</i> , a <i>Rickettsiella</i> sp., a <i>Hepatazoon</i> sp. and a <i>Bartonella</i> -like sp.  Gemmell <i>et al.</i> (1991) showed that juvenile Northern Brown Bandicoots that were infected with <i>I. tasmani</i> , <i>I. holocyclus</i> and <i>Haemaphysalis humerosa</i> were anaemic, had elevated white blood cell counts and had slower growth rates than tick-free individuals.  Spratt & Haycock (1988) found third-stage larvae of the filaroid nematode <i>Cercopithifilaria johnstoni</i> in newly moulted nymphal and adult <i>I. tasmani</i> that had recently fed on infected Bush Rats ( <i>Rattus fuscipes</i> ).
Seasonality	Not yet fully understood.

Natural Hosts	<p><b>REPTILES</b></p> <p>Widespread infestation of reptile taxa, especially Scincidae and Elapidae.</p> <p><b>MAMMALS</b></p> <p><b>Artiodactyla:</b> Bovidae, Suidae.  <b>Carnivora:</b> Canidae (including Dingo <i>Canis familiaris</i>).  <b>Dasyuromorphia:</b> Myrmecobiidae (Numbat <i>Myrmecobius fasciatus</i>).  <b>Diprotodontia:</b> Macropodidae (Eastern Grey Kangaroo <i>Macropus giganteus</i>, Kangaroo Island Western Grey Kangaroo <i>Macropus fuliginosus</i>, Barrow Island Euro <i>Macropus robustus</i>, Red Kangaroo <i>Macropus rufus</i>, Black-striped Wallaby (<i>Wallabia dorsalis</i>), Phascolarctidae, Potoroidae  <b>Lagomorpha:</b> Leporidae.  <b>Monotremata:</b> Ornithorhynchidae (Platypus <i>Ornithorhynchus anatinus</i>).  <b>Perissodactyla:</b> Equidae.  <b>Rodentia:</b> Muridae.</p>
Human Infestation	Yes.
Human Disease Risks	<p><b>BACTERIAL DISEASES</b></p> <p>Rickettsial Spotted Fever (Barrow Island, WA). Pathogen: <i>Rickettsia gravesii</i>.  Possible Lyme-like pathogens similar to those found in <i>Ixodes holocyclus</i>.</p> <p><b>VIRAL DISEASES</b></p> <p>So far, no human viruses have been isolated from <i>Amblyomma triguttatum</i>.</p> <p><b>PROTOZOAL DISEASES</b></p> <p>So far, no infectious protozoans have been isolated from <i>Amblyomma triguttatum</i>.</p>
Seasonality	Larvae are found from March to September, but more abundant in March-April. Nymphs occur from March to December with the peak number during August. Adult ticks occur from November to March with the maximum number in December-January.
<p><b>Cattle Tick</b>  <i>Rhipicephalus (Boophilus) microplus (Canestrini, 1888)</i></p>	
Known Stages	<p>Male, female, nymph, larva.</p> <p>Larvae hatch from eggs and swarm up grass blades, where they can survive up to 34 weeks before finding a suitable host. When attached to a host, they feed for about one week, moult and become nymphs. Nymphs feed on the host for a further week and moult into adults. Adult females continue feeding on the host's blood for one week before dropping into the pasture where they lay up to 3000 eggs and then die. Adult males may feed on the host occasionally, but do not fill with blood; they wander over the host for two months or more, mating with females. Eggs hatch into larvae after 2-6 months, depending on time of year.</p>
<p>Description of adult male (extract from Roberts 1970)</p> 	<p><b>Body:</b> Oval, yellowish to reddish brown in colour, 1.60 x 1.20 mm to 2.00 x 1.35 mm; expanded laterally beyond edge of scutum where it is hairless and finely striate; hairs long and numerous dorsally and ventrally but absent from grooves and depressions. Caudal appendage usually small, conical, but occasionally reduced to a mild, rounded, shallow protuberance, rarely absent. <b>Capitulum:</b> Length 0.35-0.40 mm. Basis dorsally with the lateral angles swollen, with scattered long hairs except immediately posterior to mouthparts; posterolateral margin concave, posterior margin straight or mildly convex; cornua moderate in size and usually rather blunt; basis ventrally with lateral ridges, some scattered pale bristles laterally and a pair of short, posthypostomal bristles. Palps 0.19-0.21 mm in length, hirsute; transverse ridges on articles 2 and 3 prominent, particularly in article 2 laterally; internal margin of article 2 dorsally frequently mildly indented at about mid-length; article 3 dorsally with apex flattened or sometimes rounded; both articles ventrally with a short retrograde process, that on basal article usually tongue-like, sometimes pointed; internal margin of basal article ventrally usually concave, occasionally almost straight, without any bristle. Hypostome 0.18-0.20 mm in length, broad, curved laterally; dentition 4/4 or 6-8, rarely 9, teeth per file; corona well developed. <b>Scutum:</b> Oval, the margin adjoining the spiracular plate mildly excavated; 1.60 x 1.00 mm to 1.80 x 1.19 mm. No lateral grooves; median groove elongate, moderately deep and wide; posterolateral grooves about the same length as median groove, curved convergently anteriorly; on each side and just anterior to the posterolateral grooves, a rounded depression to which these grooves sometimes extend; cervical grooves anteriorly deep and short and thence extending almost to the mid-length of the scutum as shallow and ill-defined depressions. Punctations, at most, sparse, shallow, and inapparent, distributed mainly between cervical grooves; surface with a granulated appearance, a pseudoscutum frequently faintly obvious. Eyes placed well forward, frequently difficult to detect. Scapulae strong, blunt.</p>

<b>Cattle Tick</b> <i>Rhipicephalus (Boophilus) microplus</i> (Canestrini, 1888)	
Description of adult male (extract from Roberts 1970) <i>Continued</i>	Emargination somewhat deep. <b>Genital Aperture:</b> On a level with coxa II. <b>Anal Groove:</b> Not apparent. <b>Ventral Shields:</b> Adanals somewhat variable in shape, usually subrectangular except anteriorly, three or four times as long as broad, the posterior margin variable but usually with posteroventral angle produced as a blunt or pointed mild spur; accessory shields posteriorly with the internal point blunt or pointed to varying degrees; both shields with numerous, mild punctuations and long hairs and only occasionally extending to the body margin. <b>Spiracular Plate:</b> Subcircular, greatest dimension 0.15-0.18 mm. <b>Legs:</b> Pale, increasing in length and robustness posteriorly, with many hairs dorsally and ventrally. Coxa I with an elongate, spur-like, anterior process, curved dorsad and extending well beyond the scapular, posteriorly deeply cleft, the inner spur stout and blunt, the outer spur a little smaller, more slender and more pointed; coxa II with more broadly rounded internal and external spurs, the external spur somewhat triangular; coxa III with similar but less developed spurs; coxa IV apparently without a spur, but sometimes with indications of a rounded salience. Tarsus I with a single terminal spur; tarsi II-IV each with a terminal and sub-terminal spur; length of tarsus I 0.28-0.32 mm, and of tarsus IV 0.30-0.35 mm.
Description of adult female (extract from Roberts 1970)	<div style="display: flex; align-items: center;">  <div style="margin-left: 10px;"> <p><b>Body:</b> Unfed specimens oval, narrowing behind coxa IV: 1.6 x 1.1 mm to 2.1 x 1.4 mm; dorsally and ventrally with numerous, long, pale hairs, absent from grooves; median and posterolateral grooves elongate and well defined, the latter confluent with a shallow depression on each side extending to posterolateral margins of scutum and there meeting the cervical grooves; no anal groove; median postanal groove well defined. Engorged specimens attaining 12.9 x 8.4 mm, the “waist” becoming less obvious towards full engorgement; body grooves still apparent. <b>Capitulum:</b> Length 0.45-0.55 mm. Basis dorsally with pointed lateral angles, the posterolateral angles rounded and at most only slightly salient; on each side, a longitudinal valley include the porose areas; porose areas usually oval, sometimes pyriform, moderate in size, not deep, divergent anteriorly, the interval 1-1.5 x the greatest dimension of one and sometimes a little depressed; hairs restricted to one or two in region of anterolateral margin. Palps 0.18-0.22 mm in length, not as hirsute as in male; dorsally with transverse ridges on articles 2 and 3 as in male, article 3 more pointed apically than in male; dorsal internal margin of article 2 convex and with a median indentation which is continued transversely as a mild groove; ventrally the retrograde process on articles 2 and 3 not as prominent as in male, internal margin of article 2 convex anteriorly with two or three strong bristles and of article 3 with one or two bristles basally; basal article ventrally as in male. Hypostome mildly indented apically, 0.22-0.28 mm in length; dentition 4/4 of usually eight teeth per file, but sometimes only six or as many as nine; corona of minute denticles, well defined. <b>Scutum:</b> Longer than wide, 0.81 x 0.75 mm to 1.20 x 1.00 mm, widest at level of eyes which are placed a little anterior to mid-length of scutum. Anterolateral margins a little divergent posteriorly; posterolateral margins straight or mildly curved; posterior angle rounded and relatively narrow. Punctuations not apparent, surface granular, sometimes rugose in anterior cervical and scapular fields; hairs long, sparse, absent in cervical grooves and posterior angle. Eyes oval, slightly raised, moderate in size, and usually abutting on margin or nearly so. Cervical grooves relatively narrow and convergent for a short distance anteriorly, then divergent as shallower, broader depressions to reach the scutal margins. Scapulae elongate and blunt. <b>Genital Aperture:</b> On a level with coxa II. <b>Spiracular Plate:</b> Broadly oval or subcircular, greatest dimension 0.25 mm. <b>Legs:</b> Pale and of moderate length. Coxa I triangular, anterior process not as prolonged as in male, with two broadly rounded spurs separated by a relatively deep cleft, the external spur broader than the internal spur; coxae II and III each with two shallow, broadly rounded spurs, those on coxa II being more obvious, the internal spur on coxa III sometimes difficult to detect; coxa IV with a single external spur appearing more as a salience. Tarsus I with a terminal ventral spur; tarsi II-IV also with a subterminal ventral spur; length of tarsus I 0.35-0.37 mm, and of tarsus IV 0.37-0.40 mm.</p> </div> </div>
Description of nymph (extract from Roberts 1970)	<b>Body:</b> Unfed specimens oval, 0.89 x 0.65 mm; pale, with many long, pale hairs dorsally and ventrally; the dorsal hairs erect, distributed mainly marginally and submarginally; median, posterolateral, genital, and median postanal grooves distinct. Engorged specimens attaining 2.8 x 1.7 mm, narrowing posteriorly at level of spiracular plates; all grooves still distinct. <b>Capitulum:</b> Length: 0.22-0.30 mm; cheliceral sheaths projecting beyond palpi. Basis dorsally 0.30-0.35 mm in width, about three times as wide as long, hexagonal, lateral angles usually pointed; posterolateral margins usually mildly concave, posterior margin usually mildly convex, the posterolateral angles mildly salient to form small, blunt cornua. Palps 0.10-0.11 mm in length, mildly sclerotised with few bristles; transverse ridges apparent, one bristle on ventral inner margins of articles 2 and 3. Hypostome about 0.11 mm in length, mildly indented apically and with a well defined corona; dentition 3/3 of 5 or 6 teeth. <b>Scutum:</b> About as wide as long, 0.46 x 0.45 mm to 0.51 x 0.50 mm, widest in region of eyes which are small, only mildly raised, and situated at about mid-scutal length. Anterolateral margins frequently slightly curved and almost subparallel; posterolateral margins almost straight; posterior angle relatively narrow. Surface somewhat granulated, no obvious punctuations. Cervical grooves shallow, attaining scutal margins. Hairs few and scattered. Emargination deep. <b>Spiracular Plate:</b> Subcircular, greatest dimension about 0.10 mm. <b>Legs:</b> Short and somewhat heavy, pale. Coxa I with two rounded spurs, the inner spur shallow and frequently not distinct; coxae II and III each with a single, shallow, rounded spur; coxa IV with a mild salience. Tarsi unarmed; tarsus I stout, 0.15-0.17 mm long; tarsus IV more slender 0.15-0.16 mm long.

<b>Cattle Tick</b> <i>Rhipicephalus (Boophilus) microplus</i> (Canestrini, 1888)	
Description of larva	Larvae have a short, straight capitulum and a brown to cream body. They have six legs instead of eight.
Description of eggs	No description provided.
Zoogeographic region	Afrotropical, Australasian, Nearctic, Neotropical, Oriental, Palearctic, and several oceanic islands.
Ecoregions	Many ecoregions, chiefly in tropical and sub-tropical areas worldwide.
Natural Hosts	<b>AMPHIBIANS</b> Anura: Bufonidae.  <b>REPTILES</b> <b>Squamata:</b> Chaemeleonidae, Elapidae.  <b>BIRDS</b> (several orders).  <b>MAMMALS</b> <b>Artiodactyla:</b> Bovidae (preferred hosts) (cattle, buffalo, sheep, goat), Camelidae; Cervidae; Equidae; Suidae.
Human Infestation	Yes
Human Disease Risks	In Australia, <i>Rhipicephalus microplus</i> is the only known vector of bovine babesiosis ( <i>Babesia argentina</i> and <i>B. bigemina</i> ), bovine spirochaetosis ( <i>Borrelia theileri</i> ), and of anaplasmosis ( <i>Anaplasma marginale</i> ). It is regarded as a probable source of human and bovine infection with Q Fever.
Seasonality	Each life stage can be present throughout the year, but adults and nymphs occur mainly from late spring to mid winter. Abundances on cattle increase rapidly from summer to autumn, reaching a peak on the north coast of NSW in late autumn to early winter. They decline in abundance with the onset of cold weather.
<b>Brown Dog Tick</b> <i>Rhipicephalus sanguineus</i> Latreille, 1806	
Known Stages	Male, female, nymph, larva.  The brown dog tick is a 3-host tick; this indicates that it leaves the host to develop and molt between the larval, nymphal and adult stages. Each stage must locate a host; in a domestic environment this may result in feeding on the same dog (if there is only one or a few dogs present), but there is an opportunity for the same tick to feed on three different hosts.  A fully blood-fed female brown dog tick can lay up to 5000 eggs; the number of eggs laid depends on the size of the tick and the amount of blood she ingested. The length of time each stage feeds, and the time required for development and molting, are very dependent on temperature. Feeding and development times are generally faster at warmer temperatures. Survival is generally higher at cooler temperatures and higher relative humidity, but these ticks are tolerant of a wide range in conditions.  An adult female will feed on the host for around one week, then drop off the host and find a secluded place for egg development. Cracks and crevices in houses, garages and dog runs are ideal locations. She will start laying as soon as four days after she completes feeding and drops off the host, and can continue to lay for as long as 15 days. As she lays the eggs, she passes them over her porose areas (specialized areas on the back of the basis capituli), to coat them in secretions which protect the eggs from drying out. After she finishes laying her eggs, she dies. The larvae hatch two to five weeks later, and begin to quest, or look for a host. All stages of this tick prefer dogs, although they will feed on other mammals. Larvae feed for three to seven days, then take about two weeks to develop into nymphs. The nymphs then feed for five to 10 days and again take about two weeks to develop into adults. As adults, both males and females will attach to hosts and feed, although the males only feed for short periods. The overall cycle can be completed in just over two months, but frequently will take longer if there are few hosts available or in cold temperatures. This species is long-lived, and can survive as long as three to five months in each stage without feeding.
Description of adult male (extract from Roberts 1970)	<b>Body:</b> Pear-shaped, widest in region of spiracular plates, integument bulging both laterally and posteriorly with engorgement; 2.0 x 1.5 mm to 3.3 x 1.9 mm; usually dark brown, some scattered pale hairs dorsally, more numerous and longer ventrally; festoons distinct, the median festoon usually swollen and protruding, conspicuously so in engorged specimens. <b>Capitulum:</b> Length 0.52-0.63 mm. Basis dorsally 0.50-0.63 mm in width, hexagonal, the lateral angles salient and pointed, lateral submarginal angles frequently a little swollen, surface sometimes with a few, shallow punctuations; posterior margin concave with usually strong, blunt cornua. Palps 0.22-0.29 mm in length; articles 2 and 3 subequal, article 1 seen dorsally as a sharp conspicuous, subtriangular plate, which bears on its inner margin 6-8 (usually 8) long, irregularly branched hairs; ventrally article 2 with five or six (usually five) similar hairs internally, and article three with three or four (usually three) simple hairs. Hypostome clavate, 0.26-0.30 mm in length; dentition 3/3 of seven or eight large teeth per file; corona well developed.



<b>Brown Dog Tick</b> <i>Rhipicephalus sanguineus</i> Latreille, 1806	
Description of adult male (extract from Roberts 1970) <i>Continued</i>	<p><b>Scutum:</b> Moderately convex, 1.9 x 1.1 mm to 2.9 x 1.8 mm. Lateral grooves deep, inset with large punctuations, commencing a little posterior to eyes and enclosing two festoons; median and two posterolateral grooves conspicuous, the former usually elongate oval, and the latter broadly oval to subcircular, a smaller depression on each side of midline and anterior to postlateral grooves. Punctuations consisting of (a) numerous, fairly large piliferous pits, more abundant anteriorly and including a linear arrangement on each side extending from the cervical grooves, thereafter more scattered and arranged in four more or less longitudinal rows, and (b) neumerous fine interstitial pits which vary in number, size and definition; a series of peliferous punctuations also along the lateral margins of scutum. Cervical grooves short, moderately deep anteriorly, shallow and divergent posteriorly. Eyes oval, mildly convex. <b>Genital Aperture:</b> On a level with anterior portion of coxa II. <b>Ventral Shields:</b> Adanal shields subtriangular, about 2-2.5 x as long as wide, 0.61 x 0.24 mm to 0.93 x 0.37 mm, the external and posterior margins curved, the posterior external angle broadly rounded, blunt, rarely pointed, but never spur-like. Accessory shield pointed posteriorly. <b>Spiracular Plate:</b> Narrowly elongated comma-shaped, greatest dimension 0.4-0.6 mm. <b>Legs:</b> Of moderate length and becoming stronger posteriorly. Coxa I with two long, subequal, closely set, pointed spurs, the inner broader than the outer; coxa II-IV, the posterointernal angle of coxae II and III rounded and salient, that of coxa IV with a small, pointed spur. Tarsi tapering somewhat gently; tarsus I without ventral spurs; length of tarsus I 0.57-0.61 mm, and of tarsus IV 0.50-0.55 mm.</p>
Description of adult female (extract from Roberts 1970)	 <p><b>Body:</b> Unfed specimens, oval, narrow anteriorly, 2.1 x 1.4 mm to 2.8 x 1.7 mm; marginal grooves deep, including usually two festoons; median and posterolateral grooves well developed; hairs short, pale and scattered dorsally, longer and more numerous ventrally; festoons well defined. Engorged specimens attaining 11.7 x 7.0 mm; marginal grooves not obvious, median and posterolateral grooves faint; festoons not apparent. <b>Capitulum:</b> Length: 0.60-0.70 mm. Basis dorsally 0.62-0.75 mm wide, as in male but cornua less developed; porose areas moderate in size, deep, usually oval with the long axis directed anteriorly and a little divergently, the interval more than the width of one. Palps as in male, 0.32-0.41 mm in length. Hypostome 0.35-0.39 mm in length; dentition 3/3 with eight to 10 large teeth per file. <b>Scutum:</b> Usually a little longer than wide, 1.20 x 1.20 mm to 1.60 x 1.45 mm, widest at level of eyes which are large, oval and mildly convex. Posterolateral margin obtusely angled a little posterior to its mid-length; posterior angle with a small marginal expansion. Lateral grooves frequently deep, inset with large punctuations and extending from commencement of cervical grooves to, or almost to, the scutal margin, the lateral ridge usually rounded, but sometimes carina-like. Cervical grooves short and deep anteriorly, divergent and shallow posteriorly and forming with the lateral grooves an elongate, bow-shaped depression. Punctuations moderate to numerous in number and varying in development, the larger punctuations piliferous, somewhat indiscriminately scattered but grouped mainly in scapular and cervical fields, the smaller punctuations more numerous, more evenly distributed, but frequently most numerous in median and posterior fields. Scapulae long, blunt. <b>Genital Aperture:</b> In unfed specimens on a level with coxa II, but moving to a level with first intercoxal space on engorgement. <b>Spiracular Plate:</b> Broadly comma-shaped, greatest dimension about 0.4 mm. <b>Legs:</b> More slender than in male. Coxa I as in male; coxae II-IV each with a small triangular external spur and a rounded internal salience, the spur becoming smaller posteriorly and the salience less obvious. Tarsi armed as in male, but spurs not as strong, particularly the subterminal spurs; length of tarsus I 0.60-0.70 mm, and of tarsus IV 0.52-0.60 mm.</p>
Description of nymph (extract from Roberts 1970)	 <p><b>Body:</b> Unfed specimens, oval, 0.95 x 0.55 mm to 1.15 x 0.60 mm; scutum occupying about half the length of the body; festoons well defined. Engorged specimens attaining 3.30 x 2.10 mm. <b>Capitulum:</b> Length 0.20-0.25 mm. Basis dorsally hexagonal, 0.28-0.35 mm wide, about three times as wide as long, lateral angles strongly salient and sharply pointed; posterior margin practically straight; no cornua; auriculae small and blunt. Palps about 0.15 mm in length, closely applied to mouthparts to give capitulum dorsally a pronounced triangular appearance, tapering to a blunt point, the external margin practically straight; article 1 apparent vertically as an internal lobe carrying a single, long hair branched on one side; a single internal hair on article 2 ventrally. Hypostome about 0.12 mm in length; dentition 2/2 of six or seven large teeth on each file. <b>Scutum:</b> As long as wide or almost so, 0.45 x 0.50 mm to 0.50 x 0.50 mm and widest at level of eyes which are situated posterior to scutal mid-length. Anterolateral margins practically straight; posterior angle broadly and regularly rounded. Cervical and lateral grooves much as in female, with area between lateral grooves and anterolateral margins somewhat swollen. Punctuations not obvious, surface crazed. Scapulae moderate in size, rounded. <b>Spiracular Plate:</b> Oval, greatest dimension 0.12 mm. <b>Legs:</b> Coxa I with two long subequal spurs, the external spur more slender and more pointed than the internal spur, that on coxa IV minute. Tarsi unarmed; length of tarsus I about 0.22 mm, and of tarsus IV about 0.18 mm.</p>
Description of larva	 <p>The tiny light brown larvae ("seed tick") have 6 legs and move to a lower area to attach to a dog at the first opportunity. Should a host not be readily available, they can survive for 8 months without a blood meal and water. After larvae attach to a host they engorge themselves for 3-9 days, changing from a flattened to a globular shape, and light brown to bluish-gray color. They leave the host, seek a sheltered area, and molt in 6-23 days to become an 8-legged reddish-brown nymph.</p>

<b>Brown Dog Tick</b> <i>Rhipicephalus sanguineus</i> Latreille, 1806	
Description of eggs	No description provided.
Zoogeographic region	Worldwide, but probably originated in Palaearctic.
Ecoregions	Natural range thought to be periphery areas of the Sahara and Middle Eastern deserts, but now has a worldwide distribution as a result of the domestication of dogs.  In Australia, this species finds conditions suitable for its development throughout the tropics and subtropics and especially in the drier areas, but only in man-made habitations and in association with dogs. There are rare records of this species occurring in more southern areas of Australia, e.g. Melbourne.
Natural Hosts	<b>Carnivora:</b> Canidae, mainly a parasite of the Domestic Dog.
Human Infestation	Yes
Human Disease Risks	Known globally to be a vector of Q Fever ( <i>Coxiella burnetii</i> ), Boutonneuse Fever ( <i>Rickettsia conorii</i> ) and Spotted Fever ( <i>Rickettsia rickettsii</i> ) in humans, and babesiosis ( <i>Babesia canis</i> ) and ehrlichiosis ( <i>Ehrlichia canis</i> ) in dogs.
Seasonality	Can be present throughout the year, but all life stages are more abundant during the warmer, moister months.
<b>Bandicoot Tick</b> <i>Haemaphysalis humerosa</i> Warburton & Nuttall, 1909	
Known Stages	Male, female, nymph, larva.  Life cycle not well understood, but each stage is dependent on a host to complete its development.
Description of adult male (extract from Roberts 1970)	<b>Body:</b> Markedly narrowly oval, about 2.5 x as long as wide, widest in region of first festoon; 1.8 x 0.7 mm to 2.8 x 1.1 mm. <b>Capitulum:</b> Length 0.32 – 0.50 mm. Basis dorsally subrectangular, 0.30-0.43 mm wide, not quite twice as wide as long, lateral submarginal fields a little swollen; posterior margin straight or mildly convex; cornua well developed and pointed; basis ventrally flat, broadly rounded posteriorly. Palps elongate, greatest dimension 0.30-0.42 mm, external margin practically straight; article 2 much longer than article 3, strongly salient basally, the salience directed posterolaterally, the basal margins dorsally and ventrally mildly lobed; article 3 triangular, about as long as wide, with a small, but distinct, ventral, retrograde spur. Hypostome 0.14-0.16 mm in length, clavate; dentition 4/4, rarely 5/5, of six to eight teeth; corona moderate, denticles minute. <b>Scutum:</b> Mildly convex and same shape and dimensions as body. Lateral grooves long and distinct, including usually two festoons, but occasionally one or three. Surface mildly rugose, punctuations usually numerous and mainly fine, densest medianly, with some confluency and mild ridges. Cervical grooves shallow, somewhat subparallel and extending for about 25% of scutal length. Festoons mostly longer than wide. <b>Genital Aperture:</b> On a level with coxa II. Spiracular Plate: Suboval, with somewhat flattened sides and a mild, dorsal process, about twice as long as wide, greatest dimension 0.15-0.18 mm. <b>Legs:</b> All coxae elongate and distinctly longer than wide; coxa I with strongly developed anterior prolongation which projects well beyond scapula and is readily visible dorsally, with a blunt spur of moderate length; coxa II-IV each with a single small, blunt spur. Trochanters with mild spurs. Tarsi tapering gradually, length of tarsus I 0.30-0.35 mm and of tarsus IV 0.28-0.35 mm.
Description of adult female (extract from Roberts 1970)	<b>Body:</b> Unfed specimens conspicuously elongate oval, 2.1 x 1.0 mm to 2.6 x 1.2 mm, widest in region of first festoon; marginal grooves long, deep, including two, occasionally three, festoons. Engorged specimens elongate oval, attaining 7.4 x 4.3 mm; marginal grooves not apparent; hairs few, minute, scattered. <b>Capitulum:</b> Length 0.42-0.56 mm. Basis dorsally 0.40-0.42 mm wide, slightly more than twice as wide as long, lateral submarginal areas more swollen than in male; porose areas superficial, oval, somewhat convergent anteriorly, wide apart; basis otherwise as in male. Hypostome 0.20-0.25 mm in length; dentition usually 4/4, rarely 5/5, of eight to 10 teeth; corona with minute denticles. <b>Scutum:</b> Elongate oval, almost twice as long as wide, 1.10-0.63 mm to 1.30-0.74 mm, widest at about mid-length. Punctuations moderate in number, fairly evenly distributed, mainly fine medianly and posteriorly, sometimes a little coarser laterally. Cervical grooves subparallel, moderately deep and narrow for the greater part of their length, attaining the scutal margin. <b>Genital Aperture:</b> On a level with the second intercoxal space or with coxa II. <b>Spiracular Plate:</b> Broadly suboval, greatest dimension 0.30-0.45 mm. <b>Legs:</b> Coxae as in male, but a little wider in relation to their length. Length of tarsus I 0.37-0.45 mm, and of tarsus IV 0.36-0.45 mm.
Description of nymph (extract from Roberts 1970)	<b>Body:</b> Unfed specimens elongate oval, 1.2 x 0.6 mm; marginal grooves long, deep, including two festoons. Engorged specimens elongate oval, 2.7 x 1.2 mm; marginal grooves and festoons faint. <b>Capitulum:</b> Length about 0.25 mm. Basis dorsally about 0.2 mm wide, essentially as in female. Palps as in female, but basal salience not directed posteriorly as strongly; greatest length about 0.2 mm. Hypostome about 0.14 mm in length, clavate; dentition usually 2/2 of six to eight teeth, but sometimes specimens 3/3 distally and 2/2 basally; corona with minute denticles. <b>Scutum:</b> Subcircular, 0.37 x 0.36 mm to 0.46 x 0.44 mm. Punctuations few and fine. Cervical grooves subparallel, shallow, elongate, frequently attaining scutal margin. <b>Spiracular Plate:</b> Subcircular, transverse in engorged specimens, greatest dimension about 0.11 mm. <b>Legs:</b> Coxae broader in relation to their length than in female and spurs more pointed. Length of tarsus I about 0.24 mm, and of tarsus IV about 0.21 mm.

<b>Bandicoot Tick</b> <i>Haemaphysalis humerosa</i> Warburton & Nuttall, 1909	
Zoogeographic region	Australasian.
Ecoregions	Tropical and sub-tropical grasslands, savannas and shrublands. Recorded only from the northern half of Australia. In the east, it occurs in coastal and sub-coastal areas from Iron Range, north Qld to Sydney, NSW. Its distribution in the NT appears to be confined to coastal areas and in WA to the Northwest.
Natural Hosts	Preferred hosts for larvae, nymphs and adults are bandicoots ( <b>Peramelemorphia</b> : Peramelidae).  More rarely found on:  <b>MAMMALS</b> <b>Artiodactyla</b> : Bovidae. <b>Dasyuromorphia</b> : Dasyuridae (Northern Quoll <i>Dasyurus hallucatus</i> , Yellow-footed Antechinus <i>Antechinus flavipes</i> , Atherton Antechinus <i>Antechinus godmani</i> ). <b>Diprotodontia</b> : Phalangeridae (Common Brushtail Possum <i>Trichosurus vulpecula</i> ). <b>Monotremata</b> : Tachyglossidae (Kangaroo Island Echidna <i>Tachyglossus aculeatus</i> ). <b>Peramelemorphia</b> : Peramelidae (Northern Brown Bandicoot <i>Isoodon macrourus</i> , Long-nosed Bandicoot <i>Perameles nasuta</i> , Golden Bandicoot <i>Isoodon barrowensis</i> , Barred Bandicoot <i>Perameles bougainville</i> ) <b>Perissodactyla</b> : Equidae. <b>Rodentia</b> : Muridae (Black Rat <i>Rattus rattus</i> , Bush Rat <i>Rattus fuscipes</i> , False Water Rat <i>Hydromys chrysogaster</i> , Giant White-tailed Rat <i>Uromys caudimaculatus</i> , Fawn-footed Melomys <i>Melomys cervinipes</i> , Shark Bay Mouse <i>Pseudomys praeconis</i> , Brush-tailed Rabbit Rat <i>Conilurus penicillatus</i> , Common Rock Rat <i>Zyomys argurus</i> , Pale Field Rat <i>Rattus tunneyi</i> and Black-footed Tree-rat <i>Mesembriomys gouldii</i> ).  <b>BIRDS</b> <b>Coraciiformes</b> : Coraciidae (Dollarbird <i>Eurystomus orientalis</i> ). <b>Cuculiformes</b> : Cuculidae (Pheasant Coucal <i>Centropus phasianinus</i> ).
Human Infestation	Yes.
Human Disease Risks	The same disease risks as those posed by <i>Ixodes holocyclus</i> .
Seasonality	Adults can be present throughout the year, but are most abundant during spring and summer. Larvae & nymphs least abundant during the hottest months of the year.
<b>Bush Tick</b> <i>Haemaphysalis longicornis</i> (goral) Neumann, 1901	
Known Stages	Male, female, nymph, larva.  This is a three-host tick, with each of its growing stages- larvae, nymph, and adult feeding off separate hosts, not necessarily of the same species. The adult female tick, which when fully engorged with blood can grow to approximately 9mm long by 7mm wide, lays hundreds of eggs from which the larvae will hatch on nearby vegetation. The larvae attach to a suitable host and feed before falling to the pasture where they develop to a nymph stage. Nymphs will also attach to a host to feed before detaching and developing into an adult. The time taken for the completion of the life cycle varies considerably from days to months depending on factors such as temperature and the host's immunity developed from previous exposure.  Larvae and nymphs position themselves at the tips of long grass or vegetation and attach to the skin of grazing animals or hosts walking through the paddocks.
Description of adult male (extract from Roberts 1970)	Body: Relatively broadly oval, widest at about mid-length, 2.1 x 1.5 mm to 2. X 1.7 mm; hairs minute, scattered. <b>Capitulum</b> : Length 0.46-0.56 mm. Basis dorsally about 1-1.5 x as wide as long, surface sometimes mildly punctate, lateral submarginal areas swollen; posterolateral margins straight or mildly curved, a little divergent anteriorly; posterior margin straight; cornua strong, pointed; basis ventrally flattened, wider than long, posterior margin mildly convex. Palps 0.24 x 0.27 mm in length, the contour of the external margin broken by the slightly protruding basal margin of article 3; articles 2 and 3 subequal; article 2 only moderately salient basally, the salience pointed and directed anterolaterally, the dorsobasal and ventrobasal margins mildly and regularly curved; article 3 with a strong, triangular, pointed spur a little internal to the middle of the dorsobasal margin, ventrally with a strong, triangular, pointed retrograde spur. Hypostome 0.18-0.21 mm in length, clavate; dentition 5/5, 4/4 basally, of eight to 10 teeth; corona well developed. <b>Scutum</b> : Shape and dimensions similar to those of body. Lateral grooves long, linear but well defined, may enclose the first festoon or terminate at the anterior margin of the first festoon. Punctations mainly fine, numerous, fairly evenly distributed. Cervical grooves short, not conspicuous, shallow posteriorly. Estoons mostly longer than wide. <b>Genital Aperture</b> : On a level with coxa II. Spiracular Plate: Subrectangular, with a mild rounded dorsal process; greatest dimension 0.34-0.36 mm.

<b>Bush Tick</b> <i>Haemaphysalis longicornis</i> (goral) Neumann, 1901	
Description of adult male (extract from Roberts 1970) <i>Continued</i>	<b>Legs:</b> Coxa I with a relatively long, pointed spur; coxae II-IV each with a much smaller spur, smallest on coxa IV. Trochanters each with a dark, rounded ventral spur, most distinct on I and II. Tarsi tapering gradually with a minute subapical hook; length of tarsus I 0.46-0.56 mm, and of tarsus IV 0.49-0.55 mm.
Description of adult female (extract from Roberts 1970) 	<b>Body:</b> Unfed specimens broadly oval, 2.0 x 1.5 mm to 2.6 x 1.8 mm, marginal grooves long, distinct, including one festoon; festoons about as long as wide; hairs short, pale, scattered and mainly ventral. Fully fed specimens attaining 9.8 x 8.2 mm. <b>Capitulum:</b> Length 0.56-0.65 mm. Basis dorsally 0.48-0.54 mm in width, about twice as wide as long; margins and cornua as in male; porose areas moderate in size, shallow, broadly oval to subcircular and frequently a little convergent anteriorly, wide apart, the interval frequently with a punctuate, mild depression; basis ventrally with shape as in male. Palps 0.40-0.45 mm in length, essentially as in male. Hypostome 0.30-0.37 mm in length, clavate; dentition 5/5, sometimes 4/4 basally, rarely entirely 4/4, with eight or nine teeth; corona well developed. <b>Scutum:</b> Subcircular, with mild anterolateral, posterolateral and posterior corners, 0.90 x 0.94 mm to 1.2 x 1.3 mm. Punctations mainly moderate in size, usually densest laterally, but sometimes more evenly distributed. Cervical grooves deep anteriorly, shallow posteriorly, extending for about 75% of the length of the scutum. Emargination somewhat deep. Scapulae blunt. <b>Genital Aperture:</b> On a level with coxa III in unfed specimens, but moving towards a level with coxa II on engorgement. <b>Spiracular Plate:</b> Broadly oval with a mild dorsal process; greatest dimension 0.36 mm. <b>Legs:</b> Coxae and trochanters as in male, but spur on coxa I more slender. Tarsi I up to 0.73 mm, and of tarsus IV to 0.60 mm.
Description of nymph (extract from Roberts 1970)	<b>Body:</b> Unfed specimen 1.50 x 1.03 mm, marginal grooves distinct, including one festoon. Engorged specimens attaining 2.9 x 1.9 mm. <b>Capitulum:</b> Length about 0.31 mm. Basis dorsally about 0.24 mm in width, essentially as in female; cornua distinct. Palps about 0.16 mm in length, essentially as in female, but article 3 without a basodorsal spur. Hypostome about 0.13 mm in length; dentition 3/3 of five or six teeth. <b>Scutum:</b> Subcircular and regularly rounded, 0.44 x 0.50 mm to 0.57-0.65 mm, widest a little anterior to mid-length. Punctations few, fine, scattered. Cervical grooves distinct, long, but not reaching scutal margin. <b>Spiracular Plate:</b> Shape as in female; greatest dimension about 0.18 mm. <b>Legs:</b> Coxae essentially as in female. Trochantal spurs present ventrally. Tarsi with slender apices; length of tarsus I about 0.40 mm, and of tarsus IV about 0.34 mm.
Zoogeographic region	Oriental, Oceania and Australasia. Occurs in Australia, New Zealand, Fiji and New Caledonia, China, north-eastern UUS, Korea and Japan. Opinion is divided as to whether the species is endemic to Australia or whether it was introduced accidentally from Japan.
Ecoregions	In Australia, found mostly in cattle areas of coastal NSW and south-east Qld. In NSW, it is abundant in the northern coastal watersheds, particularly between Taree and Wauchope. It occurs within the southern coastal areas to the Vic border and occasional records occur from inland areas such as Tenterfield, Mudgee, Inverell and Young. Victorian records refer to rare occurrences in the Bairnsdale to Wy Yung and Wodonga to Tallangatta areas.
Natural Hosts	<b>Artiodactyla:</b> Almost exclusively found on cattle, occasionally on sheep (Bovidae) and rarely on pigs (Suidae), goats (Bovidae), deer (Cervidae) and horses (Equidae).  Found even more rarely on:  <b>MAMMALS</b> <b>Carnivora:</b> Felidae, Canidae. <b>Diprotodontia:</b> Macropodidae (including Barrow Island Euro <i>Macropus robustus</i> , Black-striped Wallaby <i>Wallabia dorsalis</i> ). <b>Lagomorpha:</b> Leporidae. <b>Peramelemorphia:</b> Peramelidae (including Northern Brown Bandicoot <i>Isoodon macrourus</i> ).  <b>BIRDS</b> <b>Passeriformes:</b> Artamidae (Australian Magpie <i>Gymnorhina tibicen</i> ), Psittacidae (Budgerigar <i>Melopsittacus undulatus</i> ).
Human Infestation	Yes.
Human Disease Risks	<i>H. longicornis</i> is a known vector of Bovine Theileriosis ( <i>Theileria mutans</i> ) and Bovine Babesiosis ( <i>Babesia ovata</i> ) in cattle in Australia. It is suspected as a vector of Q Fever ( <i>Coxinella burnetii</i> ) and Spotted Fever ( <i>Rickettsia</i> spp.) in humans.
Seasonality	Nymphs feed in late winter and early spring, adults predominantly in mid-summer, and larvae in late summer and autumn. Unfed nymphs over-winter.  In warm, moist areas with mid winters, at least two generations can be produced each year, with larvae found in fairly large numbers in early spring as well as in summer and autumn. Adults and nymphs can be found on the host year round, except in mid-winter. In more temperate areas with more severe winters, only one generation is produced each year and hosts are free of ticks from late autumn to early spring.

	<b>Wallaby Tick</b> <i>Haemaphysalis bancrofti</i> Nuttall & Warburton 1915
Known Stages	Male, female, nymph, larva.  Life cycle not well understood, but it is known to be a three-host species.
Zoogeographic region	Australasia and possibly Indonesia.
Description of Adult Male (Roberts 1970)	<b>Body:</b> Relatively broadly oval, widest just posterior to coxa IV, 1.60 x 1.20 mm to 2.00 x 1.45 mm. <b>Capitulum:</b> Length 0.36-0.42 mm. Basis dorsally with some fine punctuations, 0.29-0.36 mm in width and about twice as wide as long, lateral submarginal fields swollen; posterolateral margin practically straight; cornua well defined, bluntly pointed; basis ventrally flattened, wider than long, the posterior margin mildly convex. Palps stout, external margin usually concave and frequently a little recurved, its contour usually broken but sometimes the posterior margin of article 3 may protrude a little, greatest length 0.25-0.30 mm; article 2 longer than article 3, strongly salient basally, the salience directed laterally, and sometimes a little anterolaterally, the external dorsobasal and ventrobasal margins lobed, the ventral lobe pronounced and sometimes bluntly angulated, the angulation occasionally elevated and spur-like, ventrally with a moderate, pointed retrograde spur. Hypostome 0.14-0.16 mm in length, clavate; dentition 4/4 of seven or eight teeth, sometimes with an additional file of two to four teeth distally; corona well defined. <b>Scutum:</b> Shape and dimensions as with body. Lateral grooves distinct, though somewhat less so in the smaller specimens, commencing on a level with cox III and including one festoon. Punctuations usually numerous, fine to moderate in size, mainly shallow, sometimes a little denser anteriorly and laterally, but in some specimens few and scattered. Cervical grooves short. Festoons mostly longer than wide. <b>Genital Aperture:</b> On a level with coxa II. <b>Spiracular Plate:</b> Suboval, a small rounded prolongation directed posterodorsally, greatest dimension 0.17-0.19 mm. <b>Legs:</b> Coxa I with a short, stout, bluntly pointed spur; coxae II-IV each with a smaller, blunt spur, smallest on coxa IV. Trochanters each with a small, blunt ventral spur. Tarsi tapering gradually, II-IV each with a minute and indistinct subapical hook; length of tarsus I 0.33-0.44 mm, and of tarsus IV 0.31-0.38 mm.
Description of Adult Female (Roberts 1970)	<b>Body:</b> Unfed specimens oval, 2.0 x 1.4 mm to 2.5 x 1.7 mm; marginal grooves long and deep, including usually two, occasionally three, festoons; festoons mostly as wide as long; hairs minute, scattered. Engorged specimens attaining 7.8 x 5.7 mm. <b>Capitulum:</b> Length 0.40-0.53 mm. Basis dorsally 0.39-0.50 mm in width and more than twice as wide as long; porose areas oval, depressed, convergent anteriorly, occasionally subcircular, wide apart; basis otherwise dorsally and ventrally as in male. Palps with greatest length 0.30-0.38 mm, essentially as in male but angulation of dorsobasal margin of article 3 usually more pronounced and more frequently spur-like. Hypostome 0.18-0.26 mm in length, clavate; dentition usually 4/4, rarely 5/5, of seven to nine teeth; corona well defined. <b>Scutum:</b> Subcircular, 0.80 x 0.79 mm to 1.03 x 1.07 mm, widest at about mid-length. Punctuations laterally usually numerous and moderate in size with confluency to give a rugose surface, elsewhere finer and more scattered. Cervical grooves long, shallow posteriorly, extending for the full length of the scutum or almost so. Emargination moderate. Scapulae blunt. <b>Genital Aperture:</b> At about a level with second intercoxal space, but moving to a level with coxa II on engorgement. <b>Spiracular Plate:</b> Broadly suboval, but occasionally as in male; greatest dimension 0.18-0.30 mm. <b>Legs:</b> Coxae and trochanters essentially as in male. Tarsi as in male; length of tarsus I 0.45-0.50 mm, and of tarsus IV 0.40-0.45 mm.
Description of Nymph (Roberts 1970)	<b>Body:</b> Unfed specimens broadly oval, 1.03 x 0.73 mm to 1.14 x 0.88 mm; marginal grooves including two festoons. Engorged specimens attaining 2.10 x 1.25 mm. <b>Capitulum:</b> Length 0.18-0.23 mm. Basis dorsally 0.20-0.23 mm in width; cornua distinct. Palps with greatest length about 0.13 mm, essentially as in female but dorsobasal margin of article 3 not angulated; articles 2 and 3 subequal. Hypostome about 0.11 mm in length; dentition 3/3, occasionally 2/2, of five or six teeth. <b>Scutum:</b> Wider than long, 0.33 x 0.44 mm to 0.43 x 0.54 mm. Punctuations few and fine, distributed mainly laterally. Cervical grooves broad and shallow, extending well into the posterior field. <b>Spiracular Plate:</b> Suboval, greatest dimension about 0.12 mm. <b>Legs:</b> Coxae essentially as in female. Trochanters each with a mild ventral spur. Length of tarsus I about 0.25 mm, and of tarsus IV about 0.23 mm.
Ecoregions	Little is known about the preferred habitat of this species. Occurs throughout coastal and subcoastal regions of Qld and northern NSW. In Qld, specimens have been collected in the Gulf of Carpentaria, Cape York Peninsula, and in numerous localities from Cairns to Brisbane and in the south-east as far west as Dalby and Gooniwindi. In NSW the species is abundant in the Northern Rivers District and occurs as far south as Nowra. It has also been recorded on Kangaroo Island in SA (Roberts 1970).  <i>H. bancrofti</i> also occurs in PNG and there is an unconfirmed record from Java, Indonesia (Barker & Walker 2014).

<b>Wallaby Tick</b> <i>Haemaphysalis bancrofti</i> Nuttall & Warburton 1915	
Natural Hosts	<p><b>MAMMALS</b></p> <p><b>Artiodactyla:</b> Bovidae (cattle, sheep), Cervidae (deer), Suidae (pigs).  <b>Carnivora:</b> Canidae (Domestic Dog <i>Canis familiaris</i> and Dingo <i>Canis lupus</i>), Felidae (Domestic Cat <i>Felis catus</i>).  <b>Chiroptera:</b> Vespertilionidae (Large Forest Bat <i>Vespadelus darlingtoni</i>).  <b>Dasyuromorphia:</b> Dasyuridae (Northern Quoll <i>Dasyurus hallucatus</i>, Spotted-tailed Quoll <i>Dasyurus maculatus</i>), Macropodidae (Agile Wallaby <i>Macropus agilis</i>, Black wallaroo <i>Macropus barnardus</i>, Swamp Wallaby <i>Wallabia bicolor</i>, Black-striped Wallaby <i>Macropus dorsalis</i>, Tammar Wallaby <i>Macropus eugenii</i>, Common Wallaroo <i>Macropus robustus</i>, Red-necked Wallaby <i>Macropus rufogriseus</i>, Brush-tailed Rock-wallaby <i>Petrogale penicillata</i>, Bridled Nailtail Wallaby <i>Onychogalea fraenata</i>, Western Grey Kangaroo <i>Macropus fuliginosus</i>, Allied Rock-wallaby <i>Petrogale assimilis</i>, Short-eared Rock-wallaby <i>Petrogale brachyotis</i>, Godman's Rock-wallaby <i>Petrogale godmani</i>, Red-necked Pademelon <i>Thylogale thetis</i>), Petauridae (Sugar Glider <i>Petaurus breviceps</i>), Phalangeridae (Common Brushtail Possum <i>Trichosurus vulpecular</i>), Phascolarctidae (Koala <i>Phascolarctos cinereus</i>), Potoroidae (Long-nosed Potoroo <i>Potorous tridactylus</i>, Rufous Bettong <i>Aepyprimmus rufescens</i>, Woylie <i>Bettongia penicillata</i>), Vombatidae (Common Wombat <i>Vombatus ursinus</i>),  <b>Peremelemorphia:</b> Peramelidae (Long-nosed Bandicoot <i>Perameles nasuta</i>, Southern Brown Bandicoot <i>Isoodon obesulus</i>, Northern Brown Bandicoot <i>Isoodon macrourus</i>) and Thylacomyidae (Bilby <i>Macrotis lagotis</i>)  <b>Rodentia:</b> Muridae (Black Rat <i>Rattus rattus</i>).</p> <p><b>BIRDS</b></p> <p><b>Cuculiformes:</b> Cuculidae (Pheasant Coucal <i>Centropus phasianus</i>).  <b>Passeriformes:</b> Artamidae (Australian Magpie <i>Cracticus tibicen</i>)</p>
Human Infestation	Yes.
Human Disease Risks	In Australia, <i>H. bancrofti</i> is essentially a parasite of macropodids, especially wallabies. However, it is frequently seen on cattle and, together with <i>H. humerosa</i> , it is one of the main vectors of bovine theileriosis ( <i>Theileria orientalis</i> ).
Seasonality	Larvae, nymphs and adults are present throughout the year. But there is no information about seasonal changes in abundances.
<b>Southern Reptile Tick</b> <i>Bothriocroton (Aponomma) hydrosauri</i> Denny 1843	
Known Stages	<p>Male, female, nymph, larva.</p> <p>Extract from Barker &amp; Walker (2014):</p> <p><i>Tiliqua rugosa</i> (Sleepy Lizard) is covered in thick overlapping scales; <i>B. hydrosauri</i> is a small tick that can easily crawl under these scales. Male and female ticks prefer to the axillae of the forelegs or in the ears of their host. Male ticks may stay attached for many months waiting for new females to attach to the host. Female ticks must take a small meal of blood 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. 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 2.0 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 8-35 days, apparently on blood, then detach to lay their eggs. Female <i>B. hydrosauri</i> lay approximately 1,500 eggs over a period as long as 41 days. Larvae attach to their host 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, they moult into 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.</p>
Zoogeographic region	Australia.

<b>Southern Reptile Tick</b> <i>Bothriocroton (Aponomma) hydrosauri</i> Denny 1843	
Description of Adult Male (Roberts 1970)	<p><b>Body:</b> Somewhat subcircular, 3.1 x 3.1 mm to 4.3 x 3.9 mm, broadest in the region of the spiracular plates; hairs small, pale numerous ventrally, sparse dorsally; five symmetrically disposed plaques ventrally, small to moderate in size, and just anterior to the festoons. <b>Capitulum:</b> Length 0.96-1.30 mm. Basis dorsally subrectangular, 0.62-0.71 mm in width, the surface with shallow punctuations; posterolateral margins mildly convex, the posterior margin almost straight or mildly concave with the posterolateral angles at most only mildly salient. Palps 0.65-0.82 mm in length, not quite four times as long as wide dorsally; article 2 about twice as long as article 3 and ventrally with an internal, distal expansion, bristles moderately numerous. Hypostome somewhat spatulate with a flattened apex and strong corona; dentition 4/4, with files of five or six strong teeth decreasing in size posteriorly and also internally, the fourth and innermost file of minute teeth irregular in disposition and number and sometimes reduced to one or two. <b>Scutum:</b> Shape and dimensions as in body; dark brown to almost black, paler medianly and posteriorly in the vicinity of the lateral grooves and sometimes with a narrow pale lateral marginal stripe. Punctuations numerous, unequal, deep, the larger punctuations distributed mainly laterally and posteriorly, the surface presenting a rugose appearance with depressions and in some specimens small, raised areas. Lateral grooves deep, with many coarse punctuations, continuous and sometimes broader and with some fine striae posteriorly. Festoons highly punctuate dorsally and ventrally, the separations deep, each festoon dark brownish medianly. Cervical grooves short, moderately deep. <b>Genital Aperture:</b> About on a level with coxa II. <b>Spiracular Plate:</b> Somewhat broadly comma-shaped, greatest dimension 0.60-0.73 mm. <b>Legs:</b> Strong, frequently dark with pale articulations. Coxa I with two short, but stout, blunt spurs; coxae II-IV each with a single, similar spur. Trochanters each with a small, dark, rounded, ventral spur. Tarsi with ventral spurs; tarsus I with three small dorsal protuberances; tarsus IV with a single, larger, dorsal protuberance; length of tarsus I 0.79-0.89 mm, and of tarsus IV 0.79-0.86 mm.</p>
Description of Adult Female (Roberts 1970)	<p>Body: Unfed specimens subcircular, 3.8 x 3.6 mm to 4.2 x 4.0 mm, thickly clothed with pale, coarse, decumbent hairs; festoons distinct. Engorged specimens subcircular to broadly oval, attaining 19.3 x 15.7 mm; festoons now not obvious. <b>Capitulum:</b> Length: 1.1-2.0 mm. Basis dorsally 0.80-1.35 mm in width; posterolateral margins convex, posterior margin sinuous and indented medianly, posterolateral angles rounded, mildly salient; porose areas large, deep, oval to subcircular, sometimes almost contiguous with posterior margin and a little divergent anteriorly, the interval about the width of one and frequently mildly depressed. Palps stout, 0.80-1.4 mm in length, article 2 essentially as in male, much longer than article 3. Hypostome with strong corona; dentition 4/4, with three files of up to eight strong teeth and a fourth innermost file of minute teeth. <b>Scutum:</b> Subtriangular and widest anteriorly, posterior angle relatively narrow and sometimes a little flattened, 1.5 x 2.1 mm to 2.6 x 3.2 mm. Brown, lighter in colour between cervical grooves, and frequently with a pale spot in the posterior angle. Punctuations numerous, unequal, mainly finer medianly with many coarse laterally, the surface frequently with a rugose appearance, some very large deep, confluent punctuations forming a conspicuous depression laterally and extending to the scutal margin just posterior to the lateral angles. Cervical grooves deep anteriorly, continuing as shallower depressions to reach the scutal margin or almost so. Emargination deep. <b>Genital Aperture:</b> On a level with coxa II and moving a little anteriorly as engorgement proceeds. <b>Spiracular Plate:</b> Broadly comma-shaped; greatest dimension 0.76-0.85 mm. <b>Legs:</b> Coxae as in male, but spurs more rounded. Trochanters each with a shallow, rounded spur. Tarsi relatively slender; ventral spurs small; dorsal protuberance not as pronounced as in male; length of tarsus I 1.0-1.4 mm, and of tarsus IV 0.84-1.22 mm.</p>
Description of Nymph (Roberts 1970)	<p><b>Body:</b> Unfed specimens dark, subcircular, 1.7 x 1.9 mm; hairs short, pale, scattered. Engorged specimens subcircular, pale, attaining 4.7 x 4.3 mm. <b>Capitulum:</b> Length 0.5-0.6 mm. Basis subrectangular, 0.35-0.40 mm in width; posterior margin straight; posterolateral angles rounded, not salient. Palps slender, 0.37-0.40 mm in length; article 2 about twice as long as article 3. Hypostome dentition 3/3 of five to seven teeth, the innermost file with minute teeth. <b>Scutum:</b> Subtriangular, much wider than long, 0.65 x 0.90 to 0.88 x 1.20 mm, the posterior angle narrow. Lateral fields with some coarse, deep punctuations, which are confluent to form a conspicuous indentation of the scutal margin just posterior to the lateral angle; punctuations otherwise small, scattered between the cervical grooves, more numerous laterally and sometimes in the posterior angle. Cervical grooves attaining posterolateral margins of scutum, deep and wide anteriorly and usually well defined for most of their length. <b>Spiracular Plate:</b> Broadly comma-shaped and almost as wide as long; greatest dimension 0.25-0.30 mm. <b>Legs:</b> Coxa I with two subequal, small, blunt spurs, II-IV each with a smaller, blunt spur. Trochanters each with a rounded ventral spur. Tarsi slender, without spurs; all tarsi with very mild protuberances; length of tarsus I 0.45-0.50 mm and of tarsus IV 0.36-0.41 mm.</p>
Ecoregions	<p>A number of studies summarised by Bull (1986) have shown that there are at least four allopatric populations of <i>B. hydrosauri</i>: (1) south-eastern SA; (2) Eyre Peninsula, SA; along the coast from Albany to Bremer Bay, WA; and the Margaret River region, along the coast from Cape Naturaliste to Cape Leeuwin, WA. Bull (1986) believes that these populations are relicts of a once much larger, perhaps trans-national, geographical distribution. Sharrad &amp; King (1981) suggest that the apparent reduction in the distribution of this species is a result of competition and range expansion of another tick species, Stumptailed Lizard Tick (<i>Amblyomma albolimbatum</i>).</p>

Natural Hosts	<p>The principal host is the Sleepy Lizard <i>Tiliqua hydrosauri</i>. However, it can be found on all the main types of reptiles that occur in southern Australia (lizards, snakes and a terrestrial turtle). When given the opportunity, it will feed on the blood of humans, cattle and horses.</p> <p>Reptile species recorded as hosts to <i>B. hydrosauri</i> include:</p> <p><b>Chelonidae:</b> Snake-necked Turtle (<i>Chelodina longicollis</i>).</p> <p><b>Agamidae:</b> Common Bearded Dragon (<i>Pogona barbata</i>), Central Bearded Dragon (<i>Pogona vitticeps</i>), Mountain Heath Dragon (<i>Rankinia diemensis</i>), Superb Dragon (<i>Diopriphora superba</i>)</p> <p><b>Scincidae:</b> White's Skink (<i>Liopholis whitti</i>), Metallic Skink (<i>Niveoscincus metallicus</i>), Southern Blue-tongue Lizard (<i>Tiliqua nigrolutea</i>), Western Blue-tongue Lizard (<i>Tiliqua occipitalis</i>), Common Blue-tongue Lizard (<i>Tiliqua scincoides</i>), Sleepy Lizard (<i>Tiliqua rugosa</i>).</p> <p><b>Varanidae:</b> Gould's Goanna (<i>Varanus gouldii</i>).</p> <p>Elapidae: Common Death Adder (<i>Acanthophis antarcticus</i>), Eastern Brown Snake (<i>Pseudonaja textilis</i>), White-lipped Snake (<i>Drysdalia coronoides</i>), Tiger Snake (<i>Notechis</i> sp.), Tiger Snake (<i>Notechis scutatus</i>), Lowlands Copper Snake (<i>Austrelaps superbus</i>).</p>
Human Infestation	Yes.
Human Disease Risks	<p><i>B. hydrosauri</i> is the arthropod host of <i>Rickettsia honei</i> on Flinders, Island, the bacterium that causes Flinders Island Spotted Fever in humans. <i>Rickettsia hoeni</i> has been isolated from the blood of patients with chronic illness, including fatigue, from Melbourne and Adelaide, but it is not known if the pathogen was the cause of the illnesses (Unsworth <i>et al.</i> 2008).</p> <p><i>B. hydrosauri</i> is also a major host and vector for <i>Hemolivia mariae</i>, a protozoan blood parasite that lives in the erythrocytes of reptiles (Smallridge &amp; Bull 1999). <i>H. mariae</i> infections appear to significantly impact on the health of lizards. There is no evidence of this parasite being transferred to humans by ticks.</p>
Seasonality	Unlike <i>I. holocyclus</i> , <i>B. hydrosauri</i> does not have a marked season of activity. However, copulation is mostly in spring and summer (September to February) when the host, mainly <i>T. rugosa</i> , is most active. Although there is some copulation in autumn and winter (March to August). Females that mate in autumn and winter delay egg-laying (i.e. enter diapause) for up to six months until the following spring (September to November) when the conditions in the soil and leaf litter are favourable for the eggs and newly hatched larvae are more likely to find a host.
<b>Seabird Soft Tick</b> <b><i>Ornithodoros capensis</i> Neumann 1901</b>	
Known Stages	<p>Egg, larva, nymph and adult.</p> <p>The life cycle is not well understood. In an experimental study, wild caught <i>O. capensis</i> fed readily on Domestic Fowl, copulation occurring shortly after feeding, and eggs first laid 45-108 days after feeding. Wild-caught nymphs moulted to the next nymphal instar (nymphal stages unknown) 32-115 days after feeding. Subsequent instars moulted to the next instar stage up to 76 days after feeding (Heath 2006).</p>
Description of adult male (extract from Roberts 1970)	<p><b>Body:</b> Variable in size, 2.5-5.0 mm x 1.4-3.0 mm; sides almost straight and subparallel for posterior 75% of length, anterolateral margins convergent to blunt, dorsal, anterior projection, broadly rounded and widest posteriorly. Dorsal and ventral surfaces with numerous, glossy, hemispherical mammillae, on dorsum larger posteriorly and laterally than anteriorly and on venter; discs dorsally large and distinct and symmetrical in arrangement, ventrally in preanal, transverse postanal, and median postanal grooves, and in three depressions posterior to postanal groove; coxal and supracoxal folds distinct. Eyes absent. <b>Capitulum:</b> Camerostome deep, bordered anteriorly by large, prominent hood and laterally by large, subrectangular cheeks separate from the hood. Basis transversely corrugated, 1.5-2.0 x as wide as long; three or four setae on each side basolaterally; posthypostomal setae short and well posterior to posthypostomal setae. Palps of moderate length; article 1 closely apposed to base of hypostome; articles 1 and 2 subequal and longer than 3 or 4. Hypostome medianly indented apically, sides subparallel; dentition 2/2 of four or five large teeth distally; some crenulations basally; corona of one or two rows of minute teeth. <b>Genital Aperture:</b> Slightly posterior to a level with coxa I. Genital apron subrectangular with rounded anterolateral angles. <b>Anus:</b> Posterior to coxa IV at about two thirds the body length. <b>Legs:</b> Relatively small, with the surface micro-mammillated. Coxa I and II separate, Coxae II-IV contiguous. Dorsal humps absent, a subapical dorsal protuberance only on tarsus I and weak; length of tarsus I 0.65-0.80 mm, and of tarsus IV 0.89-0.98 mm. No pulvilli.</p>
Description of adult female (extract from Roberts 1970)	Similar morphologically to male, 3.0-6.5 mm x 1.7-3.5 mm. General aperture a transverse slit with strong ridges radiating from slit and surrounded by a narrow-ridged tumescent margin.

<b>Seabird Soft Tick</b> <i>Ornithodoros capensis</i> Neumann 1901	
Description of nymph (extract from Roberts 1970)	Similar morphologically to the adult, but is without a genital aperture.
Zoogeographic region	Lives on the islands and occasionally, on the mainlands of the tropics and sub-tropics, of the Pacific, Atlantic and Indian Oceans between 40°N and 45°S. In Australia, it is found in various localities along the south coast, from Perth, WA to Sydney, NSW, with the Little Penguin <i>Eudyptula minor</i> as the host, and some atolls of the Coral Sea in association with the Black Noddy <i>Anous minutus</i> , Brown Booby <i>Sula leucogaster</i> and Sooty Tern <i>Sterna fuscata</i> colonies.
Ecoregions	Associated with seabird colonies. See comments for Zoogeographic Region.
Natural Hosts	Seabirds, particularly terns, gulls and penguins. However, it will feed on the blood of poultry and humans if given the opportunity.
Human Infestation	Yes.
Human Disease Risks	The tick species is likely to be responsible for the transmission of many viruses and Rickettsia spp. between birds. However, there are no confirmed cases of disease in humans caused by these pathogens. However, people who live on coral cays of the Great Barrier Reef may develop severe hypersensitivity to bites of larvae, nymphs and adults, including blistering around puncture wound, pruritis, skin lesions, rheumatic pain and lethargy.
Seasonality	Not studied. Adult, nymph and larval stages are likely to be most abundant during the nesting nest of host seabird species.
<b>Kangaroo Tick</b> <i>Ornithodoros gurneyi</i> Warburton 1926	
Known Stages	Egg, larva, between three and five nymphal instars and adult.  Life cycle described in detail by Doube (1972). All stages except the egg feed, and females may feed and oviposit as many as six times. Moulting, mating and oviposition all take place in the soil.  Adults and late-instar nymphs remain attached to their host for 20 min to 2 hrs before detaching fully engorged. In contrast, the early instar nymphs may remain attached for several days, and larvae for up to 12 days before detaching from their host fully engorged. Because kangaroos are nocturnal and range widely, feeding at night and resting under shady trees by day, there is a high probability that the engorged ticks will detach while the host is resting and so will be in place where the probability of another meal is relatively high.
Description of adult male (extract from Roberts 1970)	<b>Body:</b> Oval, subparallel and a little concave laterally, bluntly attenuated anteriorly, broadly rounded posteriorly, 4.2-5.7 mm x 3.2 mm. Dorsum with deep symmetrically disposed grooves or furrows. Mammillae numerous and crowded, mainly moderate in size, hemispherical and glossy, the surface with a few shallow pits, somewhat larger posteriorly dorsally, and towards the margins ventrally, flatter in median region. Discs arranged symmetrically, most apparent anterodorsally, otherwise disposed as a continuous series in the deep furrows. Hairs small and scattered, except along and anterolateral margins and ventrally extending to the camerostome and coxae, where they are long and curved. Coxal and supracoxal folds well developed; dorsoventral, preanal, postanal, and median postanal grooves deep. Eyes absent. <b>Capitulum:</b> Camerostome deep, camerostomal fold well developed; hood distinct, subtriangular and bent ventrad, separated from cheeks by a furrow; cheeks large, mammilated basally, and divided into a number of long finger-like processes. Basis wider than long; posthypostomal and postpalpal bristles present, the latter very short. Plaps with the surface of all articles smooth; article 1 closely apposed to mouthparts. Hypostome about 0.40 mm in length, indented apically; corona with up to five rows of two to eight minute denticles; dentition 2/2 of four or five large, pointed teeth followed by about four rows of rounded denticles which decrease in size towards the base of the hypostome, a file of some minute denticles also present internal to the files of large teeth. <b>Genital Aperture:</b> On a level with posterior edge of coxa I. Genital apron subrectangular with rounded anterolateral angles. <b>Anus:</b> In an oval frame and placed closer to the preanal than to the transverse postanal groove. <b>Legs:</b> Moderate in length with strong, long, curved hairs on legs 1 and 2. Coxae almost contiguous, with space between coxae I and II widest, a row of short, strong setae encircling the coxae and trochanters distally. All legs with prominent subapical dorsal protuberance and dorsal humps, which may be only mild on tarsus III, and are restricted to one on leg IV on the metatarsus; length of tarsus I 0.62-0.65 mm, and of tarsus IV 0.85-0.90 mm. Pulvilli absent.
Description of adult female (extract from Roberts 1970)	Similar morphologically to male. 5.6-10.0 mm x 3.2-7.2 mm. Genital aperture consists of a transverse, sometimes mildly curved, slit with narrow ridges extending to an oval, sometimes subcircular, margin.
Description of nymph (extract from Roberts 1970)	Similar to adult but lacking a genital aperture; 3.0-5.5 mm x 2.4-3.2 mm.
Zoogeographic region	Australasia.

	<b>Kangaroo Tick</b> <i>Ornithodoros gurneyi</i> Warburton 1926
Ecoregions	This species has a wide distribution in Australia and is known from open country in Qld, NSW, SA, NT and WA, particularly in areas where there are resident populations of the larger macropids and wallaroos. In Qld, specimens have been collected from the central and north-west, but there is one record of a tick being collected from a burrow in the Rockhampton area. In NSW and WA it has been seen in north-western areas, in the NT in central areas, and in semi-arid and arid areas of SA. Areas where there is a resident population of Red Kangaroo.
Natural Hosts	Principal hosts are the larger macropodids (kangaroos and wallaroos), esp. Red Kangaroo <i>Macropus rufus</i> and Common Wallaroo <i>Macropus robustus</i> . However, also known to bite humans and dogs that occur in its habitat.
Human Infestation	Yes.
Human Disease Risks	Not known.
Seasonality	<p>According to Doube (1972), most eggs are laid in spring and early summer and a few at other times of the year. There is a seasonal cycle in the incidence of imaginal disease in which most females are in diapause between mid-summer (December) and mid-winter (July). High temperatures during early summer induce diapause in the late-instar nymphs and in a proportion of the adult females. Diapause development occurs at temperatures between 10 &amp; 20 C and, in the natural environment, during autumn and winter.</p> <p>Kangaroos do not begin to spend most of the day sheltering from the heat until late spring, and so ticks are unlikely to find a meal in early spring. Even if a female tick becomes engorged in early spring, the amount of effective temperature is so small that little oogenesis occurs. In late spring and early summer (before diapauses becomes manifest), kangaroos visit wallows (a kangaroo scrape) more frequently, and temperatures become relatively high. This results in a flush of tick larvae during these periods. Kangaroos continue to visit wallows until early autumn and so the larvae give rise to a flush of first instar nymphs from January to March; these may give rise to second instar nymphs from February to April.</p> <p>January and February are the hottest and driest months of the year. Reproductive diapause in ticks halts egg production at a time when the eggs and larvae are most likely to die from desiccation.</p>

## ADDITIONAL READING

Barker, S.C and Walker, A.R. (2014). Ticks of Australia: the Species that Infest Domestic Animals and Humans. Zootaxa Monograph 3816 (Magnolia Press, Auckland, NZ).

doi: <http://dx.doi.org/10.11646/zootaxa.3816.1.1>

Bull, C.M. (1986). Ticks on reptiles, a difference of scale. Pp. 391-405. In: Sauer, J.R. and Hair, J.A. (eds). *Morphology, Physiology and Behavioural Ecology of Ticks* (Ellis Horward Limited, Chischester, New York).

Doube, B.M. (1972). The Ecology of the Kangaroo Tick *Ornithodoros gurneyi* Warburton. Unpublished Ph.D Thesis, WAITE Agricultural Institute, Adelaide.

Gemmell, R.T., Cepon, G., Green, P.E. and Stewart, N.P. (1991). Some effects of tick infestations on juvenile northern brown bandicoots (*Isodon macrourus*). *Journal of Wildlife Diseases* 27: 269-275.

Guglielmone, A.A., Robbins, R.G., Apanaskevich, D.A., Petney, T.N., Estrada-Peña, Horak, I.G. (2014). The Hard Ticks of the World (Springer, Dordrecht).

Guglielmone, A.A., Robbins, R.G., Apanaskevich, D.A., Petney, T.N., Estrada-Peña and Horak, I.G., Shao, R. and Barker, S.C.. (2010). The Argasidae, Ixodidae and Nuttalliellidae (Acari: Ixodida) of the world: a list of valid species names. *Zootaxa* 2528: 1-28.

Heath, A.C.G. (2006). Observations of *Ixodes eudyptidis* Maskell (Acari: Ixodidae), *Ornithodoros capensis* Neumann (Acar: Argasidae), and other parasites of seabirds of New Zealand. *Systematic and Applied Acarology* 11: 131-140.

Houseman, R.M. (2013). Guide to Ticks and Tick-borne Diseases (Plant Protection Programs, University of Missouri, Kansas City).

Irwin, P. (2012). Tickvectors.com website <http://www.tickvectors.com/> A research program run from Murdoch University to characterise potential pathogens in ticks across Australia.

Karl McManus Foundation website <http://www.karlmcmannusfoundation.org.au/>

Mayne, P.J. (2011). Emerging incidence of Lyme borreliosis, babesiosis, bartonellosis, and granulocytic ehrlichiosis in Australia. *International Journal of General Medicine* 4: 845-52.  
doi: [10.2147/IJGM.S27336](https://doi.org/10.2147/IJGM.S27336).

Roberts, F.H.S. (1970). Australian Ticks (CSIRO, Australia, October 1970).

Senanayake, S.N., Paparini, A., Latimer, M., Andriolo, K., Dasilva, A.J., Wilson, H., Xayavong, M.V., Collignon, P.J., Jeans, P. and Irwin, P.J. (2012). First report of human babesiosis in Australia. *Medical Journal of Australia* 196(5): 350-352. doi: [10.5694/mja11.11378](https://doi.org/10.5694/mja11.11378).

Sharrad, R.D. and King, D.R. (1981). The geographical distribution of reptile ticks in Western Australia. *Australian Journal of Zoology* 29: 861-873.

Smallridge, C.J. and Bull, C.M. (1999). Transmission of the blood parasite *Hemolivia mariae* between its lizard and tick hosts. *Parasitology Research* 85: 858-863.

Spratt, D.M. and Haycock, P. (1988). Aspects of the life history of *Cercopithfiilaria johnstoni* (Nematoda: Filarioidea). *International Journal for Parasitology* 18: 1087-1092.

Unsworth, N., Graves, S., Nguyen, C., Kemp, G., Graham, J. and Stenos, J. (2008). Markers of exposure to spotted fever rickettsiae in patients with chronic illness, including fatigue, in two Australian populations. *Quarterly Journal of Medicine* 101: 269-274.

## THE PLATYPUS AND THE ENVIRONMENTAL IMPACT ASSESSMENT PROCESS: MORE COGITATIONS OF A CONSULTANT.

Tom Grant  
Adjunct Senior Lecturer  
School of Biological, Earth and Environmental Sciences  
UNSW  
and Consultant  
Education and Environment Services Pty. Ltd.

### INTRODUCTION

The Action Plan for Australian Mammals 2012 (Woinarski, Burbidge and Harrison, 2014) has proposed that the platypus be assigned a conservation status of 'near threatened'<sup>1</sup>, due to documented population declines in some places, especially in Victoria. This downgrade may in fact be welcome to some extent, as the species' previous status - 'least concern' (IUCN) or not listed (EPBC and state threatened species schedules<sup>2</sup>) - has meant it is often

given scant consideration in development projects and their environmental assessment - "don't worry about the platypus, it isn't a threatened species and there are plenty of them around".

Being small, cryptic, mobile, hard to catch, aquatic, fossorial and mainly nocturnal, it is very difficult to assess its occurrence and numbers, and monitoring studies are problematic. In a previous article in *Consulting Ecology* (Grant, 2009a) and in Grant (2012), I described the current methods used in attempts to monitor population numbers (capture and observations) and pointed out that, especially where populations are small (and resultant capture and/or observations rates are low), it may not be possible to design a monitoring program that is capable of detecting the presence or absence of detrimental effects of a development project. Also, being a long-lived species (up to 21 years in the wild; Grant, 2004a), long-term impacts may not be quickly detected, particularly when monitoring is based on observations, rather than captures<sup>3</sup>. This is especially so where natural perturbations, such as drought and flood, may confound the interpretation of monitoring data. For this reason, and because this is a species absolutely dependent on inland water bodies and their riparian margins for its food, shelter and reproduction (Grant, 2007a), it is imperative that maximum consideration be given to minimising or eliminating any predicted adverse impacts BEFORE proceeding with any development, rather than depending on a monitoring program to detect problems down the track, which may or may not be able to be detected or addressed.

In this article, I take the opportunity to provide some suggestions, methods and protocols that may facilitate prediction and minimisation of deleterious effects of projects on the platypus, which involve disturbance of the instream and/or riparian margins of inland waters

---

<sup>1</sup>**NEAR THREATENED (NT)** □ A taxon is Near Threatened when it has been evaluated against the criteria but does not qualify for Critically Endangered, Endangered or Vulnerable now, but is close to qualifying for or is likely to qualify for a threatened category in the near future.

<sup>2</sup>In South Australia it is listed as 'endangered', as it now apparently only occurs naturally in sections of the Glenelg River in the east of the state, where the river crosses the Victorian border. There is also an introduced population at the western end of Kangaroo Island.

<sup>3</sup>Captures of lactating females in the breeding season and/or newly-emerged juveniles in late summer-autumn can give some indication of recruitment, while similar numbers seen during the monitoring from year to year may not represent a successful breeding population (Grant 2009a)

(e.g. agricultural practices, forestry, mining and construction and operation of bridges, pumping stations, dams, weirs, culverts, pipelines). These are not 'rocket science'. Some may already be known/applied by consultants and other approaches may be as good or better. However, I attempt here to draw on four decades of working with the platypus, both in research and consulting, to make some hopefully useful contribution to the species' protection and conservation in a time of rampant development and exploitation of our environment.

## IDENTIFICATION OF PLATYPUS OCCURRENCE AT A SITE

The flow chart in Figure 1 outlines a protocol for helping decide if the platypus occurs at a site.

A couple of examples. As part of the intermittent monitoring of water transfers from the Shoalhaven River into the storage dams in the Hawkesbury-Nepean River system by the Sydney Catchment Authority (SCA), three sites on the lower Wingecaribee River were netted for a mark-release-recapture study of platypuses in that system (Grant, 2006). On four

sampling occasions, with similar levels of flows and the same netting effort, the three sites were also observed on one or two occasions in the two hours following first light (never on the morning immediately following a night netting session at that site). Figure 2 shows the numbers captured and observed for each site. The numbers seen and captured were typically quite variable and numbers captured were, with two exceptions significantly higher than those observed (means over the three sites; 3.7 observed [n = 18] and 5.6 captured [n = 12];  $p < 0.05$ ).

Fortunately in the Wingecaribee River study, platypuses were actually captured at each netting session and only a single observation session yielded zero sightings. However, in other areas platypuses can be observed but not caught and caught but not seen. Two intensive observational surveys were conducted in the upper Georges River and yielded no verified platypus sightings (Grant, 2002; Grant, Durman and Durman, 2008). More recently however, one was found dead in an abandoned yabby trap, two others drowned after becoming tangled in fishing line in the river near Campbelltown (Gorrey, 2011; Partridge, 2014) and several verified sightings have been made in the River near Campbelltown in the past 12 months (personal observation).

**Given the species' ability to remain cryptic and/or not readily be captured, it should probably be assumed that the platypus could be present if the site is within the recorded distribution and the habitat is suitable (see PLATYPUS HABITAT REQUIRMENTS BELOW).**

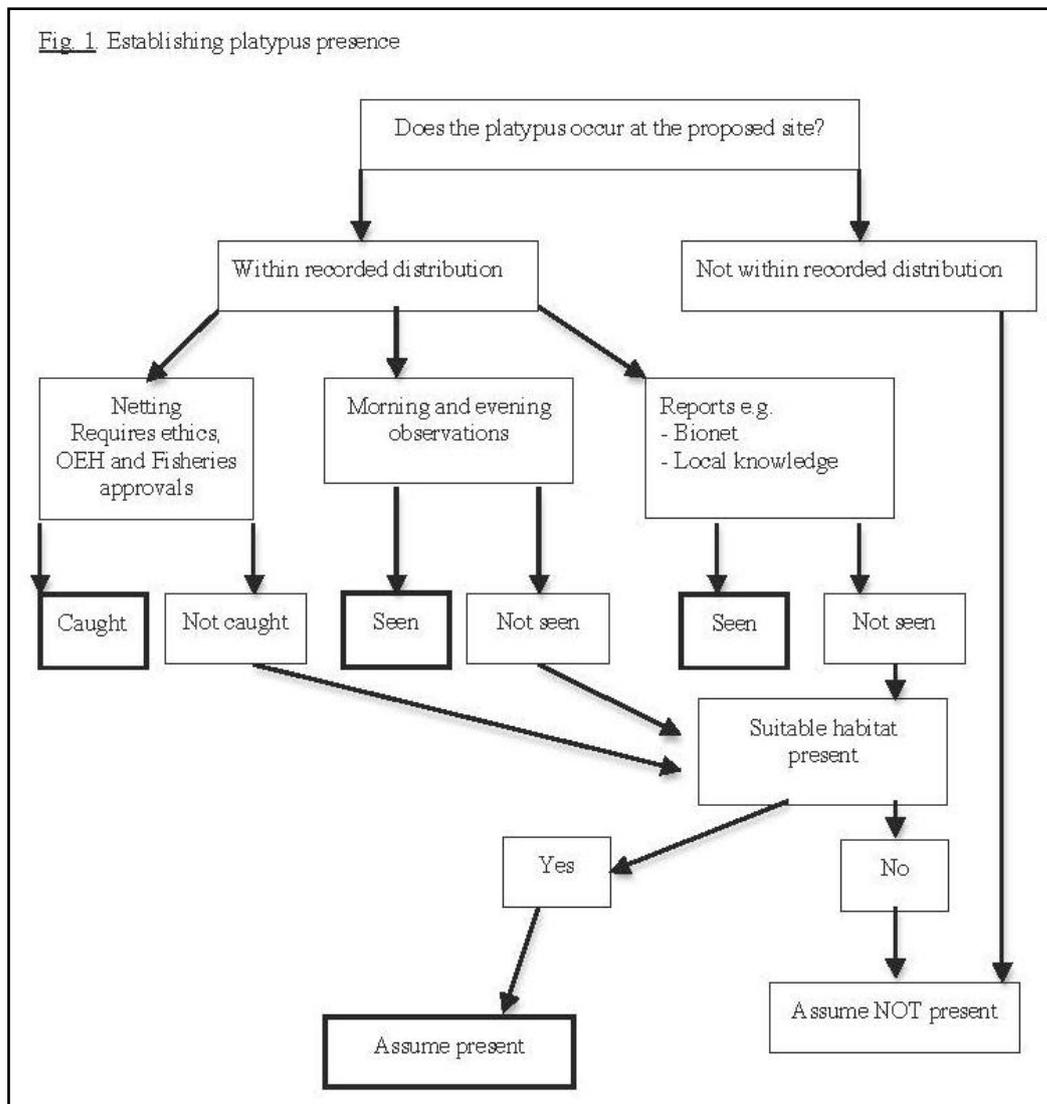
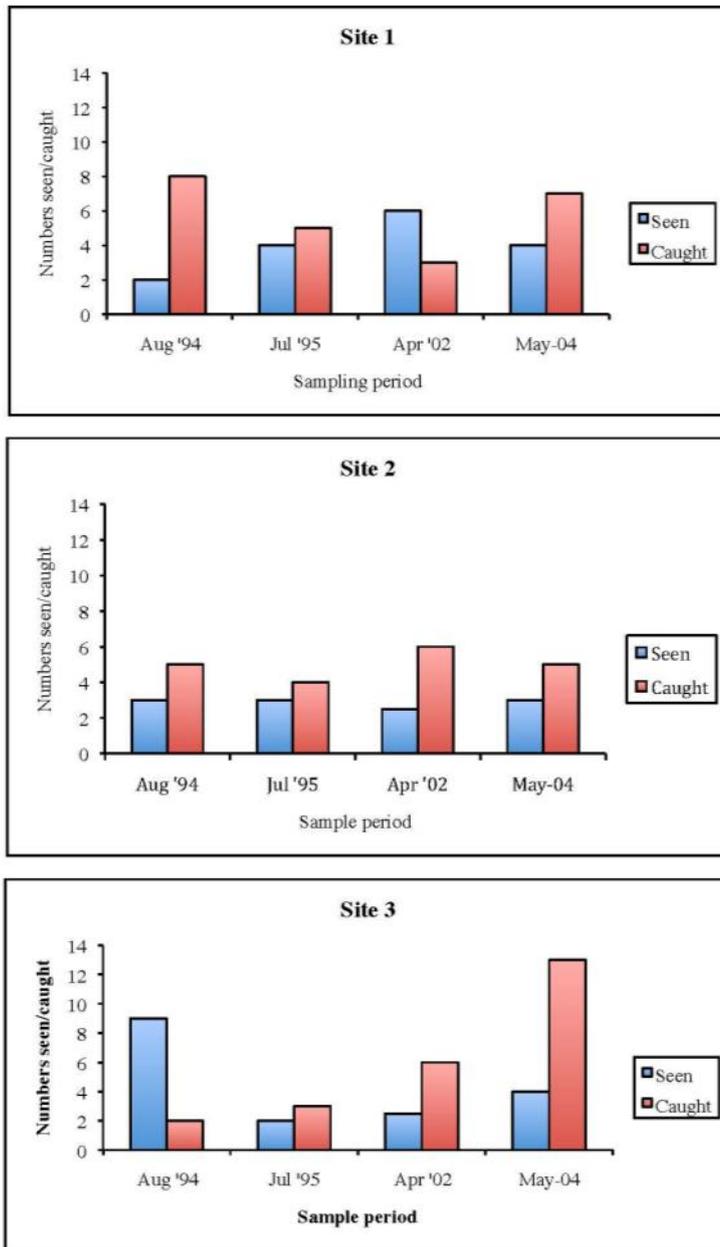


Fig. 2 Numbers of platypuses seen and captured at three sites on the Wingecarribee River



### THE BEST TIME TO DETECT PLATYPUS OCCURRENCE

Platypuses can be seen in the water at any time of the day but are normally actively foraging during the night, although quite a few emerge within the two hours before dark and may stay out foraging for a similar time after first light.

*Are morning or evening observations the best to detect presence or to get an indication of numbers?*

Some observers have found more occurrences (presence/absence in an observation session) and/or higher numbers seen per session/transect in the morning, others the evening and yet others reported no difference. Recent acoustic tracking studies found differences between sexes, and/or seasons and/or locations in diurnal activities, including morning

versus late afternoon/evening (Griffiths, Kelly and Weeks, 2014 and Josh Griffiths pers.com.) My recommendation is for BOTH early morning and late afternoon/evening to be used.

Over a number of years we carried out observations from kayaks (4 x 1 or 1.5km transects) in the two hours immediately after first light and the two before dark in the lower Manning and Hastings Rivers, and found significantly more positive sightings (occurrence per session/transect) and higher numbers (individuals seen<sup>4</sup> per session/transect) in the hour immediately before dark or that after first light than in the later hour in the morning or earlier hour in the evening.

*What about season of the year?*

Platypuses tend to spend more time in the water during the breeding season (looking for mates, collecting nesting material, consuming more food during lactation) (Grant 2007a). As a result, if the timing allows, observations from July to October in NSW and Victoria are possibly a little more likely to detect the presence or some idea of abundance than at other times of the year (Grant, 1983, 2008; Easton, Serena and Williams, 2013). However, platypuses are often seen at other times of the year.

It was expected that numbers of platypuses seen per kayak transect would be higher in late

February in NSW, when juveniles should have just left the nesting burrows and not yet dispersed (Grant 2007a; Williams, Serena and Grant, 2013). But, with the platypus you must always expect the unexpected. In two years of surveying the sites on the lower Manning and Hastings Rivers, we found that numbers per transect were not different, or in some instances lower, in February sampling periods compared to September/October.

<sup>4</sup>Numbers of individuals seen. Within the same pool multiple individuals should only be counted as different individuals if a) they are seen on the surface at the same time or b) one has just dived as another surfaced. Where pools are very long or a transect is through several pools, there needs to be some professional discretion by the observer. For example, a platypus seen leading into a riffle of one pool, a platypus seen at the top end of the next pool would probably be counted as a single individual. On the other hand, a platypus seen diving at one end of a pool, say > 50 long, a further sighting within less than about a minute (normal dive time are between 0.5 and 1.5 minutes) at the opposite end of the pool should probably be considered as a second individual.

## PLATYPUS HABITAT REQUIREMENTS

So what represents “good” or even “suitable” platypus habitat?

*“Soon the river was before me, the banks of which were adorned by pendulous Acacias, which at this season of the year [September] were profusely covered with their rich golden and fragrant blossoms, while the lofty majestic Eucalypti or Gum-trees, many of which were young and gracefully pendent, together with the Swamp Oaks or Casuarinaea, resembling firs at a distance, added to the variety and natural beauty of the landscape”. ... “The sun was now near its setting, when, at a more quiet part of the river (knowing, as I did, the crepuscular nature of the animals) I endeavoured to obtain a sight of the shy Ornithorhynchus paradoxus”. .... “At a tranquil part of the river, called by the colonists a ‘pond’, on the surface of which numerous aquatic plants were growing profusely, or in places of this description, the Water-Moles were most commonly seen, seeking their food among the plants, whilst the shaded banks afforded them the excellent situations for excavating their burrows”.*

A 'platypus heaven' is a river or stream, much like that described above by the early naturalist George Bennett (1860) in the 1830s. Important aspects are earth banks, consolidated by the roots of native vegetation, with the foliage of this vegetation at shrub and tree height providing shading of the stream and cover near the bank, where platypuses may forage and where they enter and leave both nesting and resting burrows. Many stream habitat features influence the abundance of macroinvertebrate species, which are the major food resource of the platypus. These include the presence of logs, twigs, roots and instream vegetation, as well as the type of benthic substrate. Higher macroinvertebrate productivity is usually associated with areas where logs, roots and vegetation provide a range of habitats and food for these prey species and where a cobbled or gravel substrate provides heterogeneous fixed habitat, rather than shifting substrates, such as sand. The complexity of benthic habitats and the presence of aquatic vegetation have also been identified as indicators of the occurrence of platypuses, which feed both in pools and riffles (Grant, 2007a, McLachlan-Troup, Dickman and Grant, 2010). The presence of pool-riffle sequences is also often associated with the occurrence of the species.

Grant and Bishop (1998) discussed habitat variables, usually associated with the occurrence of the platypus. Later studies also identified some of the same habitat

variables as being important and added others. These are given in Table 1, with notes on how these relate to the biology of the platypus. Associated with several consulting projects, I later used these variables to develop a scoring system as a method for rapid assessment of the habitat quality of streams and their riparian zones, with regard to potential suitability for occupation by platypuses and/or to assess how proposed developments might impact on platypuses as a result of changes in one or more of the habitat variables used in the scoring (e.g. Grant 2007b, 2009b). This scoring methodology has been used to compare streams within or between catchments, or as a comparison of separate reaches of an individual stream, but is not considered applicable to make a comparison between small parts of a single reach.

Table 2 shows a scoring system, where various important habitat variables are used to score the proportion of a stream reach exhibiting a particular variable. For example, where it is estimated that large-to medium-sized trees with roots consolidating the banks are present on greater than 75% of the stream bank (including both sides) a score of 4 would be given for that variable. Low overhanging vegetation gives the stream shade and a source of organic input, as well as acting to permit platypuses to forage safely near the banks (Table 1). In a case where only around 20% of banks in the reach have low overhanging vegetation, a score of only 1 would be given. As shown in the table, the variables are separated into those related to the stream margin (Riparian) and those of the stream itself (Stream and Substrate). In Table 2, there are 8 Riparian variables (a maximum potential reach score  $4 \times 8 = 32$ ) and 4 Stream and Substrate variables (maximum score  $4 \times 4 = 16$ ). Together these give a maximum potential score for the reach of 48 ( $32 + 16$ ). The higher the total score the more suitable the reach is thought to be for use by the platypus. Scores out of 48 in some instances are more conveniently presented as percentage figures (e.g.  $42/48 = 87.5\%$ ).

Until recently, no such scoring system had been empirically tested against either platypus occurrence or abundance. However, using mean catch per unit effort (CPUE<sup>5</sup>) data and a measure of reproductive success<sup>6</sup> at 29 locations on streams around Melbourne, Griffiths *et al* (2014) found a positive relationship between both of these variables and habitat scores

<sup>5</sup>CPUE = Platypuses captured per net night with 1 net night = 1 fyke net set at a site for a single night.

<sup>6</sup>Reproductive Success = number of juveniles captured per adult caught at a site

**Table 1. Habitat variables identified as being related to the occurrence of platypuses.**

<b>Habitat Variable</b>	<b>Known or Potential Effects on the Platypus</b>
<b><i>Bank Characteristics</i></b>	
Consolidated bank	Maintenance of shelter and nesting burrows
Bank vegetation (especially native)	Shade, bank consolidation, shelter while foraging, organic input to stream ecosystem
Large-medium sized trees on bank	
Overhanging vegetation	
Earth bank	Construction of shelter and nesting burrows
Bank height >1 metre	Construction and maintenance of shelter and nesting burrows
Absence of bank erosion	Construction and maintenance of shelter and nesting burrows; maintenance of riparian vegetation; reduction of sedimentation
<b><i>Water Body Characteristics</i></b>	
Pool depth (<2-5 metres but >1 metre)	Energetic demand of foraging increases with depth; risk of predation in shallow water
Large woody debris (LWD)	Habitat for macroinvertebrate prey species
Pool length	Foraging habitat availability
Coarse organic matter	Habitat and food for macroinvertebrate prey species
Coarser benthic substrates	
Absence of sand accumulation	
Absence of silt/clay	
Presence of macrophytes	

**Table 2. Example of a data sheet used to score habitat suitability of a stream or stream reach.**

[Scores: 0 = 0%; 1 = up to 25%; 2 = up to 50%; 3 = up to 75%; 4 = up to 100%]

<b>Habitat Variable</b>	<b>Reach Description:</b>		
1. Consolidating bank vegetation		Temperature:	Visibility:
2. Large-medium trees on bank			Flow:
3. Overhanging vegetation [ $\leq$ 1-2 metre]			
4. Overhanging vegetation [ $>$ 2 metre]			
5. Earth bank			
6. Bank height >1 metre			
7. Concave/Vertical bank			
8. Absence of bank erosion			
<b>Riparian Total (max = 32)</b>			
1. Depth >1 metre			
2. Large woody debris (LWD)			
3. Coarse organic matter (SWD/litter)			
4. Rough substrate - cobbles/gravel/pebbles			
<b>Stream and Substrate Total [max = 16]</b>			
<b>GRAND TOTAL (max = 48)</b>			
- Macrophytes			
- Sand/Mud/Bedrock			
- Connectivity			
- Burrow entrances			
- Platypuses seen [S], reported [R], caught [C]			
<b>Photographs:</b>			
<b>Grid Reference:</b>			

Variables below GRAND TOTAL (max = 48) are noted but not scored

(CPUE  $r^2 = 0.167$   $p = 0.028$ ; Reproductive success  $r^2 = 0.387$   $p = 0.01$ ). Habitat scoring was carried out over 50m stream sections, with at least five at each location (~1km apart). At 21 sites across four different waterways, scoring was carried out by two independent assessors. Although some individual scores varied, there was close correspondence of overall scores for each location (Spearman's Rank Correlation  $R = 0.889$   $p \leq 0.001$ ).

The authors of this report concluded that habitat scores represent a useful measure of habitat quality and "may help to predict the suitability of a waterway or area to support a platypus population". However, they stress the spread in their data, with some higher scores not corresponding with higher CPUE or reproductive success and vice versa.

Other, less easily quantifiable, habitat variables are probably important, including stream connectivity and isolation of populations or groups within a population. Natural perturbations (e.g. drought, flood, bushfires) also need to be considered, as these can affect habitat variables in the short- and/or long-term, depending on timing and severity. For example during a drought, parts of a stream may lose connectivity, or even experience population declines, and despite returning to a high habitat score, not continue to support a platypus population. Refinement and further testing of habitat scoring systems needs to be done.

**It should be noted that** the utilisation of a scoring system, using habitat variables known to be associated with the presence (or common occurrence) of the platypus, must be done cautiously. Such a scoring system **DOES NOT NECESSARILY:**

- **indicate that platypuses will definitely (or even more often) be found in streams or stream reaches with high scores and not (or less often) be found in streams with low scores.**
- **give any indication of probable platypus abundance.**

However, field data suggest that such a scoring system **DOES** indicate:

- **A higher probability of platypuses occurring in or using a stream or reach with a higher score than one with a lower score.**
- **The possibility that the numbers of platypuses**

**occurring in or using a stream or reach with a higher score would be greater than one with a lower score.**

It has also been popularly suggested that the presence of the platypus is an indicator of high water and/or habitat quality. There is little or no support for this contention, as platypuses are found in both pristine streams and in quite degraded ones (Grant, 2007a).

*So why use a habitat scoring system at all?*

Often projects are not fixed in terms of site, location or even the size of the area to be affected. Using habitat scores to rank stream reaches, or possible alternative streams, can inform decisions with regard to minimizing possible impacts of a development on the platypus. For example, constructing a crossing, bridge or trenching a pipeline through a stream may have high potential impact on platypus foraging, resting and nesting areas in a reach where earth banks are well consolidated by riparian trees, there is an abundance of low overhanging shrubs and the substrate consists of cobbles and gravel (in other words a reach with a high habitat score). On the other hand it may be possible to carry out this development in a lower scoring reach, where sandy banks, with little riparian vegetation cover and a sandy stream substrate occurs. The latter alternative is likely to have a much lower potential impact on platypuses in the stream.

Also, constructing and completing a scoring sheet focuses the consultant's attention on carefully assessing the stream, rather than simply giving it a general description. Critically, as shown on the scoring sheet in Table 2, there needs to be a place for notes and a list of other factors that may be important (e.g. pool connectivity, presence of burrow entrances – see *PLATYPUS BURROWS* below).

When assessing projects restricted to a specific stream reach, a habitat score may be of little help in giving advice, but the description and other factors may better inform advice to minimize impact. For example, adding another lane to an existing bridge or river crossing. The bank immediately upstream of the existing structure is consolidated with roots of trees and shrubs, is mainly earth, greater than a metre in height and exhibits burrow entrances (high riparian habitat score). On the downstream side the riparian margin consists mainly of a rocky platform, a sandy bank consolidated only by kikuyu grass and *Lantana* sp and has no apparent burrow entrances (lower riparian

habitat score). Cobbles and gravel substrates with woody debris and litter occur in other sections of the reach and in other pools upstream and downstream. It is of little consequence that the substrate immediately upstream of the existing structure is almost exclusively sand with little woody material or litter (low instream habitat score), while a mixture of gravel and sand occurs downstream and there is a bit more woody debris and litter (higher instream habitat score) adjacent to the existing structure. Literature tells us that platypuses are mobile, often foraging several kilometres over a single feeding session (Grant, 2007a; Serena and William, 2013), so that the riparian variables would be the most important in this instance. As a result advice/recommendation of the consultant would be that adding the extra lane on the upstream side would be more detrimental to the local platypus population than its construction on the downstream side of the existing structure.

### PLATYPUSES AND WILLOWS

Serena, Worley, Swinnerton and Williams (2001) reported that radio-tracked platypuses spent less time in stream reaches where willows (*Salix* sp) were the dominant riparian vegetation. The effects and management of willows is complex and controversial (e.g. Zukowski and Gawne, 2006) but platypuses are frequently found in streams where willows occur and even where they may be the dominant riparian tree species. For example, 76% of radio-tracked platypuses in the upper Shoalhaven River had their resting burrows under established willow trees, within a narrow strip of riparian vegetation, including eucalypts, wattles and tea-tree (Grant, 2007a). With regard to platypus habitat requirements, willows certainly do consolidate stream banks, although banks consolidated by established native species normally exhibit more of the bank features in Table 2 than banks

where willow predominate.

### PLATYPUS BURROWS

When I have been asked “is this a platypus burrow”, either in the field or in a photo sent by email, most often I have to say simply that “I don’t know”.

Lots of animals dig holes in banks and more than one species use the same holes, although not usually at the same time. The early naturalist Harry Burrell (1927) described the burrow tunnel as being “low-arched above and flat below – and measuring four to six inches in width [10-15cm], and from three to four inches in height [7.5-10cm], corresponding pretty well accurately to the cross section of the occupant” (Table 3). He also indicated, as I have found, that the actual entrance at the surface of the bank may be much larger and not the typical “low-arched” shape, presumably due to erosion by flows and possible further excavation by other animals (e.g. eastern water dragon, *Physignathus lueurii*). Platypuses and water rat (*Hydromys chrysogaster*) have also been recorded as using each other’s burrows at different times (Gardner and Serena, 1995). Burrell (1927) further indicated that no material comes out of the entrance of the burrow when the platypus is digging and that the soil is packed against the walls of the tunnel by the bill and tail, so that ‘fresh soil’ outside a burrow entrance probably does not indicate a platypus has been burrowing, as I have seen stated in at least one consultant’s report. Platypus faeces (which are very black, smelly and looking a bit like a wood duck scat) are semi-solid and normally voided in the water. But, for some reason in Tasmania, platypus scats are sometimes deposited near burrow entrances. Apart from a single report, I have not seen or heard of this on the mainland.

Table 3. Some useful observations/dimensions\* relating to platypus burrows

Burrow characteristic	Resting Burrow	Nesting Burrow	Comment	Reference
Tunnel width (cm)	10-15	10-15	Flat bottom curved roof	Burrell, 1927
Tunnel height (cm)	8-10	8-10		
Depth below surface (cm)	31-46	mostly 31-46 ≤120	Sloping up from entrance. May be deeper to avoid obstacles	
Rest/nest chamber-water edge (m)	0.5-3.7	mainly 2-18	Tunnels to nesting chambers usually sinuous rather than straight	Burrell, 1927 Serena <i>et al</i> , 1998
Mean	1.5±0.9	≤30		
Bank height at burrow chamber (m)	0.5-3.0	-		Serena <i>et al</i> , 1998
Mean	1.8±0.8			
Entrance height above normal water level (m)	0.1 to variable	Variable ≤12	Often under undercut banks. Occasionally under water surface	Burrell, 1927 Serena <i>et al</i> , 1998

\* Burrell (1927) mainly relating to nesting burrows on northern tablelands of NSW. Serena, et al (1998) measurements from strongest radio signal from tracked platypuses in burrows along several streams around Melbourne.

In general, unless confirmed by an observation or remote camera of a platypus entering or leaving, the presence of a burrow entrance does not necessarily indicate platypus occurrence, although a tunnel behind the entrance with a “low-arched” shape and around dimensions given above, would be more likely than not indicate a platypus burrow. Nonetheless, burrow entrances of any sort within a site to be disturbed must be further investigated for possible use by the platypus or other species that might be affected by disturbance (see *PROJECTS INVOLVING DISTURBANCE TO WATER BODIES AND/OR THEIR BANKS* below and Appendix A.).

Burrow entrances can be at normal stream level, often under overhanging banks, even numbers of metres from the stream edge or under the water surface and so are often difficult or impossible to find (Burrell, 1927; Serena, Thomas, Williams and Officer, 1998).

*Resting burrows.* These burrows are normally short (<5m; Table 3.) and simple, consisting of a single tunnel leading to a resting chamber. Radio-tracking studies have shown that platypuses will use more than one resting burrow, even during a single foraging session (Grant, 1983; Grant, Grigg, Baird and Augee, 1992; Serena, 1994, Gardner and Serena, 1995, Serena *et al*, 2002). Disturbance by construction activities could result in mortality or injury to any platypus actually being in a burrow disturbed by excavation, pile driving or sealing with rock fill and/or geotextile. However, it can probably be assumed that any platypus displaced from a resting burrow by construction activities will utilize one or more other resting burrows, rather than being permanently displaced from the area.

*Nesting burrows.* These on the other hand can be branched and complex in structure, possibly with several excavated chambers. Although often reported as sinuous in nature, they can be up to 30m long, although reported as being less 15m (Table 3). These may be refurbished or modified over several breeding seasons (Burrell, 1927, Hawkins and Battaglia, 2009). They may represent a critical habitat resource and need to be protected if possible. Also, if newly-laid eggs or dependent young (up to 3) are within a nesting burrow which is damaged or covered during construction activity, there is the potential for mortality of up to four individuals (including the lactating mother). Such an outcome can be avoided completely by construction activity being carried out outside the breeding season. The earliest a lactating (meaning she has dependent young in the burrow) female has been found in NSW is

late September and the latest mid-March (Grant, Griffiths and Temple-Smith, 2004), with lactation lasting around 4 months (Hawkins and Battaglia, 2009; Grant *et al*, 2004). It should be recommended that bank disturbance that could impact on potential nesting burrow sites should be avoided by restricting the activity to between mid-March and late-September. Unfortunately around 6 months is a long time within the life of a project. However, as most dependent young in NSW (and probably Victoria; Williams, Serena and Grant, 2012), are housed within nesting burrows between November and the end of February, the risk of mortality of dependent young in nesting burrows increases from mid-September to be very high in November, December, January and February, decreasing through to mid-March. This risk assessment is summarised in Figure 3. The chances of an institution with expertise in rearing nestlings exposed by excavation up until around January is minimal, with the possibility for successful captive rearing increasing as the nestlings become more developed from mid-January to mid-March. Any excavation activities in the breeding season should be accompanied by prior contact with such an institution (Taronga Zoo in Sydney or Healesville Sanctuary in Victoria).

Knowledge of the dimensions of platypus resting and nesting burrows relies mainly on observations made by early naturalists (especially Bennett, 1860 and Burrell, 1927), who excavated these either in an attempt to study their characteristics and/or to collect eggs or nestlings. There has been little research on the nature of either resting or nesting burrows since then, except for a radio-tracking study by Serena *et al* (1998). For this reason, any burrows or burrow complexes unearthed during excavations should be described, mapped, measured and photographed (see Appendix A.)

## PLATYPUS FOOD

Platypuses forage almost exclusively on benthic macroinvertebrate animals (Faragher, Grant and Carrick, 1979; McLachlan-Troup *et al*, 2010) and so are potentially impacted by activities that may disturb the substrate of a stream or water body, including deposition of fine sediment. As discussed above (*PLATYPUS HABITAT*), disturbance of riparian vegetation and/or woody instream material can also impact on platypus shelter, reproduction and stream productivity.

## PROJECTS INVOLVING DISTURBANCE TO WATER BODIES AND/OR THEIR BANKS

Built into any planning documentation for a project, involving disturbance to a water body or its riparian fringes (e.g. Construction Environment Management Plan), should be a number of mandatory precautions to protect the platypus and other wildlife using the stream and/or its riparian margins. Where possible these include:

- Restricting activities to the already most disturbed sections of bank, avoiding reaches with high habitat scores or those otherwise assessed as representing good platypus habitat
- Maintaining riparian vegetation and/or revegetation with local indigenous species
- Maintaining and/or realigning large woody material existing within the stream, around any existing structures or along the riparian fringes
- Maintaining industry standard sediment and pollution control, including planning for extreme rainfall events.
- Design and construction of any instream structures (e.g. bridges, culverts) to meet NSW Fisheries requirements.

Where banks are assessed as being suitable for occupation by platypus resting or nesting burrows, any excavation within 50m of the edge of the water body (Table 3) should be kept to the minimum area required and be carried out slowly and carefully in the following steps:

1. Clear vegetation around the excavation site.
2. Search for burrow entrances and carefully investigate any entrances/tunnels found by hand and/or by small excavator (using a non-toothed excavator bucket) in order to capture<sup>7</sup> and release or allow the escape of any burrow occupants.
3. Even if no burrow entrances are found<sup>8</sup>, excavation should involve relatively small scoops (10-20 cm deep), using a non-toothed excavator bucket (e.g. mud or batter bucket).
4. Constant checking for any burrows/tunnels and carefully excavating as in 2. Above.

5. As there is very little current knowledge of the nature of platypus burrows, any burrow or burrow complex found should be photographed, measured and described as in the example data sheet in Appendix A. This information should be made available to a relevant scientific institution, where it can be archived (e.g. university, museum, Office of Environment and Heritage in NSW) for future reference.

Figure 4. summarises risk assessment and precautions of bank excavation outside and within the recognised platypus breeding season in New South Wales. An example of a stepwise protocol for the supervision of such activities by an on-site ecologist/environmental officer/consultant is provided in Appendix A.

## SOURCE INFORMATION, CITATION AND SCIENTIFIC LITERATURE.

When it comes to assessment of possible impacts developments on animals, plants and the environment in which they occur, there are two well-known phenomena with which biologists are familiar but are less well understood by engineers, the proponent, commissioners and judges:

- *all predictions of impacts (or the lack of them) are based on probability, not certainty*
- *one should always expect the unexpected.*

For these reasons it is imperative that a consultant be familiar with recent findings related to the organisms under consideration. It is not acceptable to rely on *Google* or *Wikipedia* and it is also not acceptable to use the information or inferences from unpublished previous reports of other consultants, unless they have been subject to some peer review. Here is an example of how dubious conclusions can be made by not consulting the peer-reviewed literature:

In an assessment of impacts of the then current water resource development in a Queensland river, the consultant suggested that “platypus[es] feed by

<sup>7</sup> Any platypus that needs to be captured must be picked up by the tail, assuming it to be a male with spurs, and as quickly as possible be released into the water body adjacent to the development site. Where possible this should be done by a trained animal ecologist.

<sup>8</sup> Serena *et al* (1998) located 57 burrows occupied by radio-tracked platypuses but could only find the entrances to 6 of these (10.5%)

Figure 3. Risk assessment matrix for construction involving bank excavation. The boxes of animals indicate greater risk.

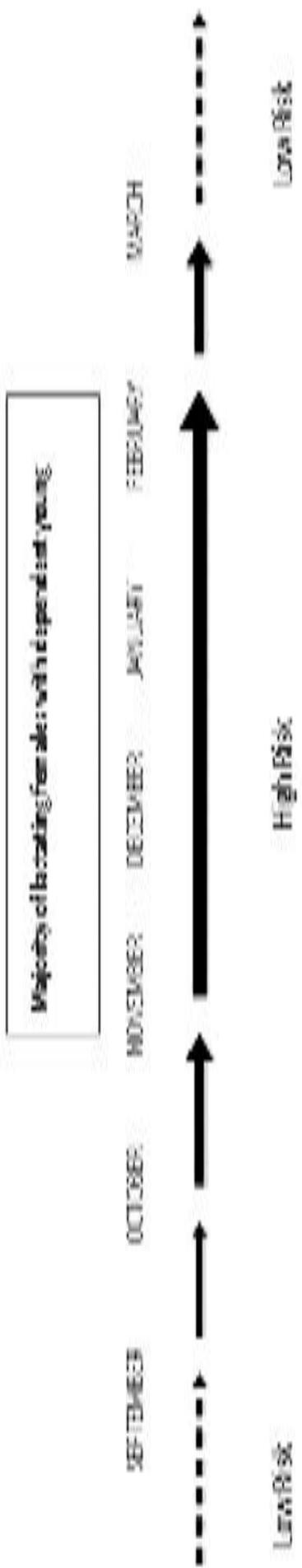
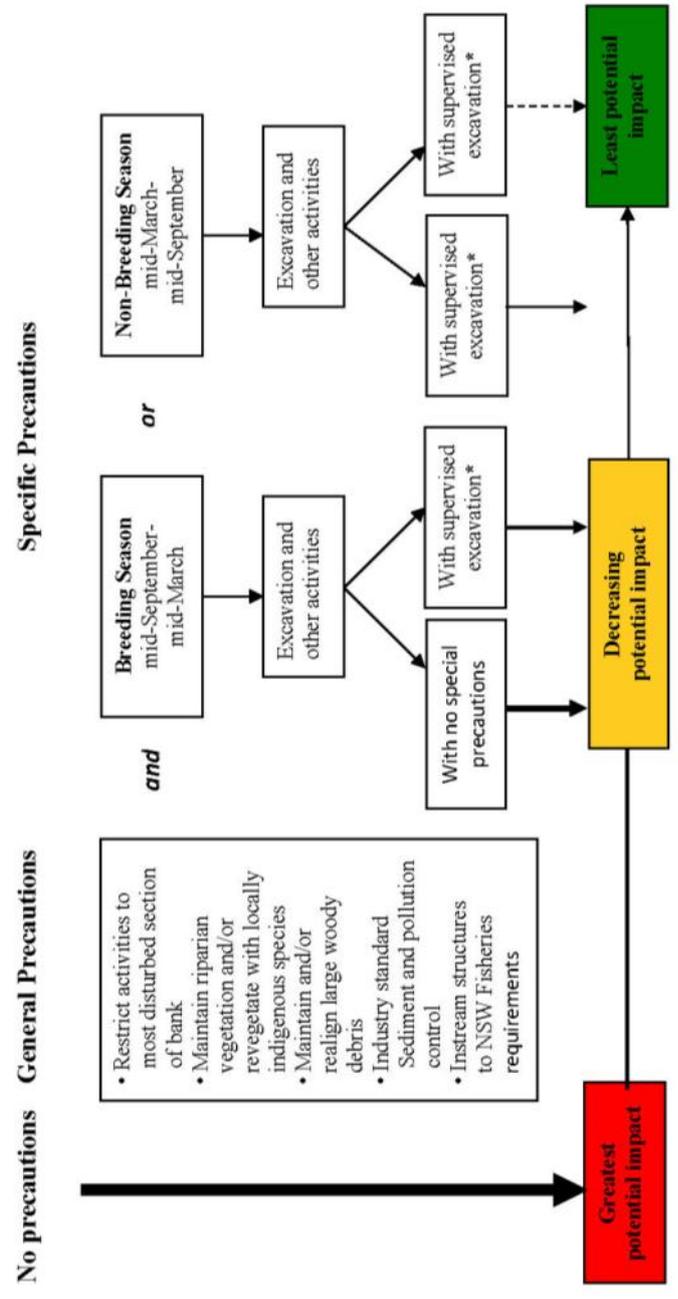


Figure 4. Flow diagram outlining the recommendations and graded associated risk of impacts on platypuses of activities involving stream bank disturbance:



\* Capture and release of adult animals affected by excavation and/or translocation of dependent young to a zoo/sanctuary with platypus care facilities

scooping prey items and mud into cheek pouches in the mouth and grinding the mixture to a sludge before digesting it". Based on this suggestion, the conclusion was that "the deposition of sediments in the shallower areas of the dam would provide extra foraging area for Platypus" as "an increase of available muddy substrate would provide more foraging area. Hard substrates offer less feeding opportunities because prey cannot be as easily scooped and ground up if they are on hard substrates or if the scooped material contains large pebble material (Anon. 2003<sup>9</sup>)."

Scientific literature was available at the time (and more is now available e.g. Grant 2004; McLachlan-Troup *et al*, 2010) that showed in fact that platypuses exhibit a preference for foraging on coarser substrates (Table 1.), which are also known to be more productive than fine sediments when it comes to the macroinvertebrate food of the platypus.

I have included some of my own unpublished experiences and quoted from several consultant reports. However, in order to stress the importance of the point made above, I have tried as much as possible in this article to provide available peer-reviewed literature to support most of its content.

## REFERENCES

- Bennett, G. 1860. *Gatherings of a Naturalist in Australasia*. John Van Voorst, Paternoster Row. London. Facsimile Edition Currawong Press. Sydney. 1982.
- Burrell, H. 1927. *The Platypus*. Angus and Robertson. Sydney.
- Easton, L, Williams, G and Serena, M. 2008. Monthly variation in observed activity of the platypus *Ornithorhynchus anatinus*. *Victorian Naturalist* **125**, 104-109.
- Dawson, T.J., Grant, T.R. and Fanning, D. 1979. Standard metabolism of monotremes and the evolution of homeothermy. *Australian Journal of Zoology* **27**: 511-515.
- Gardner, J.L. and Serena, M. 1995. Observations on activity patterns, population and den characteristics of the water rat, *Hydromys chrysogaster* along Badger Creek, Victoria. *Australian Mammalogy* **18**, 71-75.
- Gorrey, M. 2011. Second platypus found, *Campbelltown and Macarthur Advertiser*, 09 November 2011
- Grant, T.R. 1983a. The behavioural ecology of monotremes. in Eisenberg, J.F. and Kleiman, D.G. (eds.). *Advances in the Study of Mammalian Behavior*. Special Publication #7, American Society of Mammalogists.
- Grant, T.R. 1983b. Body temperature of free-ranging platypuses, *Ornithorhynchus anatinus*, with observations on their use of burrows. *Australian Journal of Zoology* **31**: 117-122.
- Grant, T.R. 1992. Captures, movements and dispersal of platypuses, *Ornithorhynchus anatinus*, in the Shoalhaven River, New South Wales, with evaluation of capture and marking techniques. in. *Platypus and Echidnas* (M.L. Augée, ed.). Royal Zoological Society of NSW, Sydney. Pp. 255-262.
- Grant, T.R. 2002. Platypus (*Ornithorhynchus anatinus*) survey of the upper Georges River, New South Wales. Report to National Parks Association of NSW. Macarthur Branch. June 2002
- Grant, T.R. 2004a. Captures, capture mortality, age and sex ratios of platypuses, *Ornithorhynchus anatinus*, during studies over 30 years in the upper Shoalhaven River in New South Wales. *Proceedings of the Linnean Society of NSW* **125**, 217-226.
- Grant, T.R. 2004b. Depth and substrate selection by platypuses, *Ornithorhynchus anatinus*, in the lower Hastings River, New South Wales. *Proceedings of the Linnean Society of NSW* **125**, 235-242.
- Grant, T.R. 2006. Platypus studies in the Wingecarribee and upper Nepean River systems between 1991 and 2006, including periods of extended operational water transfers during 2003-2006. Report prepared for Sydney Catchment Authority. June 2006.
- Grant, T. 2007a. *Platypus*. 4th Edition. CSIRO Publishing, Collingwood, Victoria.
- Grant, T.R. 2007b. Proposed augmentation of Chaffey Dam: Environmental Assessment: The Platypus. Report to GHD Services Pty. Ltd. and NSW State Water August 2007.

---

<sup>9</sup>Another consultant report to the proponent.

- Grant, T.R. 2008. The Hastings District Water Supply Augmentation Scheme: Detection of Potential Future Water-Extraction Impacts on the Aquatic Biota of the Lower Hastings River. Monitoring Study: The Platypus. Ten Year Progress Report Winter 1998 to Spring 2007. Progress Report to October 2007 prepared for K. Bishop on behalf of NSW Department of Public Works and Hastings Council. Progress Report. January 2008.
- Grant T. 2009a. The platypus and the environmental impact assessment process: Some cogitations of a consultant *Consulting Ecology*: Newsletter of the Ecological Consultants Association of NSW **23**: 50-58.
- Grant 2009b. Review of Environmental Factors for the Mardi to Mangrove Link Project. Vol. 3 Appendix G Platypus Assessment. GHD. June 2009.
- Grant, T. 2012 Environmental impact assessment: monitoring from a platypus perspective. Pp. 107-113 in *Science under siege: zoology under threat*, edited by Peter Banks, Daniel Lunney and Chris Dickman. Royal Zoological Society of New South Wales, Mosman, NSW, Australia.
- Grant, T.R., Grigg, G.C., Beard, L.A. and Augee, M.L. 1992. Movements and burrow use by platypuses, *Ornithorhynchus anatinus*, in the Thredbo River, New South Wales. in. *Platypus and Echidnas* (M.L. Augee, ed.). Royal Zoological Society of NSW, Sydney. Pp. 263-267.
- Grant, T.R., Durman, P. and Durman, B. 2008. Platypus (*Ornithorhynchus anatinus*) Survey in Stokes and O'Hares Creeks, upper Georges River Catchment, New South Wales. Report to Department of Conservation and Climate Change. June 2008.
- Grant, T.R., Griffiths, M. and Temple-Smith, P.D. 2004. Breeding in a free-ranging population of platypuses, *Ornithorhynchus anatinus*, in the upper Shoalhaven River in New South Wales - a 27 year study. *Proceedings of the Linnean Society of NSW* **125**, 227-234.
- Griffiths, J., Kelly, T. and Weeks, A. 2014. *Impacts of high flows on platypus movements and habitat use in an urban stream*. (Report to Melbourne Water). cesar, Parkville. June 2014.
- Griffiths, J., Kelly, T. van Rooyen, A. and Weeks, A. 2014. *Distribution and relative abundance of platypuses in the greater Melbourne area: survey results 2013/14*. (Report to Melbourne Water). cesar, Parkville. July 2014.
- Hawkins, M and Battaglia, A. 2009. Breeding behaviour of the platypus (*Ornithorhynchus anatinus*) in captivity. *Australian Journal of Zoology* **57**, 283-293
- Lunney, D., Grant, T. and Matthews, A. 2004. Distribution of the platypus in the Bellinger River catchment from community knowledge and field survey and its relationship to river disturbance. *Proceedings of the Linnean Society of NSW* **125**, 243-258.
- McLachlan-Troup, T.A., Dickman, C.R. and Grant, T.R. 2010. Diet and dietary selectivity of the platypus in relation to season, sex and macroinvertebrate assemblages. *Journal of Zoology* **280** (3), 237-246.
- Serena M. & Williams G.A. (2013) Movements and cumulative range size of the platypus (*Ornithorhynchus anatinus*) inferred from mark-recapture studies. *Australian Journal of Zoology* **60**, 352-359.
- Serena, M., Thomas, J.L., Williams, G.A. and Officer, R.C.E. 1998. Use of stream and river habitats by the platypus, *Ornithorhynchus anatinus*, in an urban fringe environment. *Australian Journal of Zoology* **46**, 267-282
- Serena, M., Worley, M., Swinnerton, M. and Williams, G.A. 2001. Effect of food availability and habitat on the distribution of platypus (*Ornithorhynchus anatinus*) foraging activity. *Australian Journal of Zoology* **49**, 263-277.
- Williams GA, Serena M and Grant TR. 2013. Age-related change in spurs and spur sheaths of the platypus (*Ornithorhynchus anatinus*). *Australian Mammalogy* **35**, 107-114.
- Woinarski J.C.Z., Burbidge A.A. & Harrison P.L. 2014. *The Action Plan for Australian Mammals 2012*. CSIRO Publishing, Melbourne.
- Zukowski, S. and Gawne, B. 2006 Potential Effects of Willow (*Salix* spp.) Removal on Freshwater Ecosystem Dynamics. A Literature Review. A report prepared for the North East Catchment Management Authority. Murray-Darling Freshwater Research Centre.

<sup>1</sup>To be avoided if possible between late-September and mid-March

<sup>2</sup>See attached burrow measurement table.

# APPENDIX A. PLATYPUS - RECOMMENDED PROTOCOLS FOR CONSTRUCTION ACTIVITIES INCLUDING STREAM BANK WORKS, BRIDGES, CULVERTS AND STREAM AND POND WORK - ROLE OF ON-SITE BIOLOGIST/ECOLOGIST / ENVIRONMENTAL OFFICER

## A. Pre-activity survey for potential nesting burrows:

- Activity outside breeding season (mid-March to late September) - **go to B.**
- Activity inside breeding season <sup>1</sup>-> **contact platypus specialist.**

## B. Supervision of excavation, and monitoring, by appropriately trained biologist:

Type of activity:

- Excavation in ephemeral drainage line – **got to 1.0**
- Excavation in permanent tributary stream – **got to 2.0**
- Bridge construction – **go to 3.0**
- Micro tunnel under stream– **go to 4.0**
- Other excavations of banks – **go to 5.0**

### 1.0 Excavation in ephemeral drainage line

- 1.1 Stream should be dry during construction.
  - 1.2 Check significant rain events are not forecast for construction period.
  - 1.3 Check that sediment and spill protection measures are in place.
  - 1.4 Check for any burrow entrances:
    - none present -> **no further action**
    - entrances found -> **follow careful excavation procedures:**
      - Excavation with 10-20 cm scoops with non-toothed bucket.
      - Check for the presence of voids/burrows during excavation.
      - Investigate, photograph and measure<sup>2</sup> any burrows as excavation proceeds.
- Capture and release any platypus encountered into nearest permanent stream reach.

### 2.0 Excavation in permanent tributary stream

- 2.1 Stream should be at low flows during construction.
- 2.2 Check significant rain events are not forecast for construction period.

- 2.3 Check that sediment and spill protection measures are in place
- 2.4 Check for suitable burrowing banks and/or presence of burrow entrances
  - neither present -> **no further action**
  - suitable bank and/or entrances found -> **follow careful excavation procedures:**

- Excavation with 10-20 cm scoops with non-toothed bucket.
- Check for the presence of voids/burrows during excavation.
- Investigate, photograph and measure<sup>3</sup> any burrows as excavation proceeds.
- Capture and release any platypus encountered into nearest permanent stream reach.

### 3.0 Bridge construction

- 3.1 Where possible bridge abutments should be outside riparian zone.
- 3.2 Check significant rain events are not forecast for construction period.
- 3.3 Check that sediment and spill protection measures are in place.
- 5.4 Check for any proposed disturbance in the riparian zone

- none -> **no further action**
- any excavation -> **follow careful excavation procedures:**

- Excavation with 10-20 cm scoops with non-toothed bucket.
- Check for the presence of voids/burrows during excavation.
- Investigate, photograph and measure<sup>3</sup> any burrows as excavation proceeds.
- Capture and release any platypus encountered into nearest permanent stream reach.

### 4.0 Micro tunnel under stream

- 4.1 Check significant rain events are not forecast for construction period.

---

<sup>1</sup>To be avoided if possible between late-September and mid-March

<sup>2</sup>See attached burrow measurement table.

<sup>3</sup> See attached burrow measurement table.

4.2 Check that sediment and spill protection measures are in place around boring site and spoil storage area.

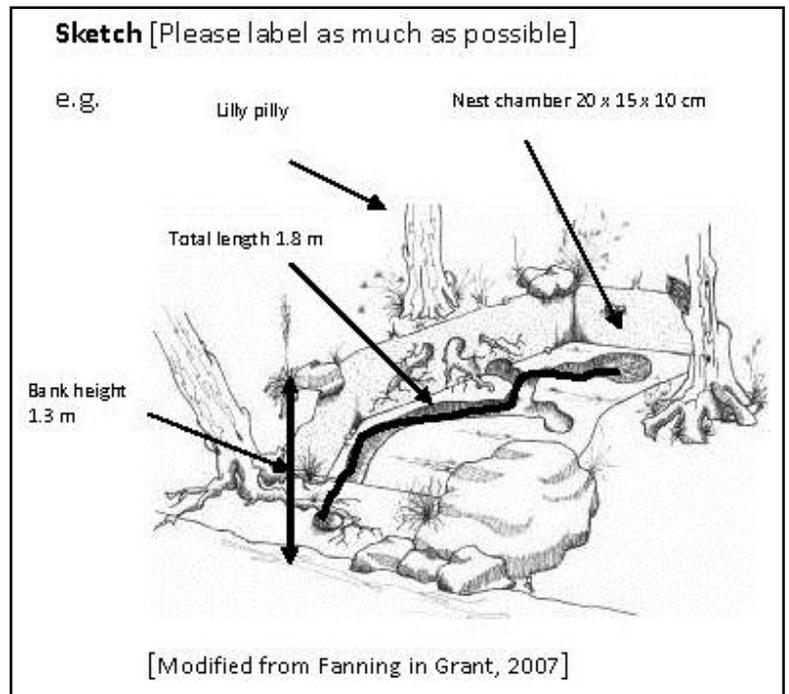
4.3 Assess possible vibration/noise levels in riparian zone

- likely to be minimal in banks where burrows may occur - **go to 4.4**
- concern that levels may be high -> **contact platypus specialist.**

4.4 Check for any proposed disturbance in the riparian zone

- none -> **no further action**
- any excavation -> **follow careful excavation procedures:**

- Excavation with 10-20 cm scoops with non-toothed bucket.
- Check for the presence of voids/burrows during excavation.
- Investigate, photograph and measure<sup>3</sup> any burrows as excavation proceeds.
- Capture and release any platypus encountered into nearest permanent stream reach.



## 5.0 Other excavation of banks

5.1 Check that sediment and spill protection measures are in place.

5.2 **Follow careful excavation procedures:**

- Excavation with 10-20 cm scoops with non-toothed bucket.
- Check for the presence of voids/burrows during excavation.

- Investigate, photograph and measure<sup>3</sup> any burrows as excavation proceeds.
- Capture and release any platypus encountered into nearest permanent stream reach.

Burrow measurements:	
Structure	Attributes/measurements
Main tunnel length [entrance to end] (metres or cm)	
Side branch length(s) (metres or cm)	
Tunnel cross sections [width x height] (cm) [Please sketch cross section profile]	
Dimensions of chambers(s) if present [length x width x height] (cm)	
Nesting material present?	
Number of entrances	
Location of entrance(s) [e.g. height above water level, distance from water edge]	
Other: [e.g. bank height, soil type, riparian vegetation around burrow]	
Details of photographs taken [Please provide tape measure or object for scale]	

The ECA Council meet every three months to discuss and deal with any current business of the association. The last meeting took place on the 8th of September and the next meeting is planned for the 8th of December. Any member who wishes to view the minutes from any of the ECA council meetings may do so by contacting the Administration Assistant Amy Rowles [admin@ecansw.org.au](mailto:admin@ecansw.org.au)

## Contributions to the Newsletter, Volume 34

Contributions to the next newsletter should be forwarded to the administration assistant Amy Rowles [admin@ecansw.org.au](mailto:admin@ecansw.org.au) by the

15th of January 2015.

- Articles may be emailed in WORD, with photos included or referenced in an attached file as a jpg.
- Please keep file size to a minimum, however there is no limit on article size (within reason)
- Ensure all photos are owned by you, or you have permission from the owner
- Ensure that any data presented is yours and you have permission from your client to refer to a specific site (if not please generalize the location).
- All articles will be reviewed by the editorial committee, and we reserve the right to request amendments to submitted articles or not to publish.
- Please avoid inflammatory comments about specific persons or entity

The following contributions are welcome and encouraged:

- ◇ Relevant articles
- ◇ Anecdotal ecological observations
- ◇ Hints and information
- ◇ Upcoming events
- ◇ Recent literature
- ◇ New publications (including reviews)
- ◇ Photographs

## Advertising Opportunities with the ECA

### Website:

\$200 for a banner

\$300 for company name with some detail and a link

\$500 for company name within box, logo, details and web link

*All website packages run for one financial year and include a small ad in any newsletter produced during the financial year.*

### Newsletter:

\$100 for a third of a page

\$250 for a half page

\$500 for a full page

\$1 / insert / pamphlet

*Advertising is available to service providers of the Ecological Consulting industry. The ECA will not advertise a consultant or their consulting business.*

If you wish to advertise, please contact the ECA administrative assistant on [admin@ecansw.org.au](mailto:admin@ecansw.org.au).

# ECA Photo Gallery

## Photo Competition Entries



**Left:** Gouldian Finch (*photo courtesy of Steve Sass*).

**Right:** Koala at Dubbo (*photo courtesy of Phil Cameron*)



**Right:** *Grevillea parviflora* subsp *parviflora* (*Photo courtesy of Isaac Mamott*).



**Below left:** Chestnut-breasted Mannikin (*Photo courtesy of Steve Sass*).

**Below centre:** Bush-stone Curlew in South-east Queensland (*Photo courtesy of Phil Cameron and Ady Watson*)

**Below :** Spiny-cheeked Honeyeater (*Photo courtesy of Steve Sass*).



# ECA Photo Gallery

## Photo Competition Entries



**Left:** Gouldian Finch (*photo courtesy of Steve Sass*).

**Above:** *Varanus rosenbergi* (*Photo courtesy of Steve Sass*)

**Below left:** Crimson Rosella (*Photo courtesy of Steve Sass*)

**Below:** Peregrine Falcons at Nymagee (*Photo courtesy of Phil Cameron*)

