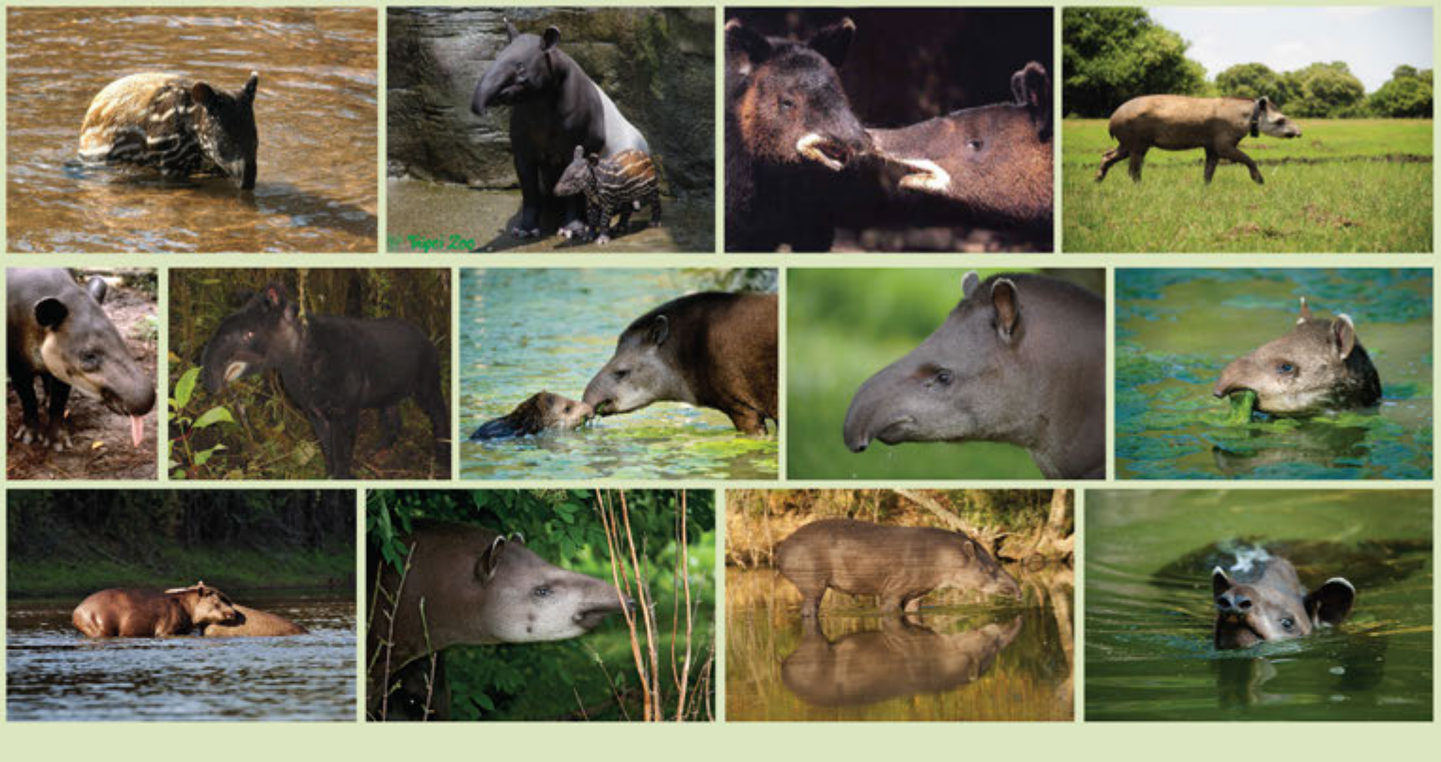

TAPIR VETERINARY MANUAL

2nd Edition | 2014

Viviana Quse & Renata Carolina Fernandes-Santos

IUCN/SSC
TAPIR SPECIALIST GROUP (TSG)
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INTRODUCTION

This is an updated version of the first Tapir Veterinary Manual published by the IUCN/SSC Tapir Specialist Group (TSG) in 2007.

Several veterinarians, biologists, nutritionists, reproduction physiologists and researchers reviewed and made contributions to this document based on their in-situ and/or ex-situ experience with the four tapir species.

The 13 Chapters and several appendices in this manual offer valuable information on many important topics for veterinarians working with tapirs, including: handling tapirs in the wild and in captivity, anesthesia protocols, treatment protocols and guidelines for medical and nutritional care.

Our hope is that this text will be helpful to all professionals working with tapir species around the world, and will contribute to the conservation of tapirs and their remaining habitats.

Editors

TAPIR SPECIALIST GROUP

IUCN SSC Species Survival Commission

Picture: Daniel Zupanc

1

Tapir Health and Conservation Medicine

Tapir Health and Conservation Medicine

Wild populations of many animal species are declining at an alarming rate. In some cases, species have disappeared without the scientific community being able to adequately learn about their basic natural history, ecology, physiology or behavior. Several species have had their conservation efforts severely threatened by the occurrence of disease epidemics, and over the past decades, health issues have become a concern to those professionals working with wildlife. Such is the case for tapirs. Population and Habitat Viability Assessments (PHVAs) carried outby the IUCN/SSC Tapir Specialist Group (TSG) have listed health issues, particularly infectious diseases and effect of toxic substance as potential threats to maintain health populations and for the survival and persistence of all tapir species in the wild (Hernandez-Divers et al. 2005; Medici et al.2007, Mangini et al. 2012).

The deforestation and habitat fragmentation associated with the conversion of tapir habitat to agricultural lands are perhaps the main drivers of disease epidemics and threats to tapir health (Medici2010). These activities result in increasing contact between tapirs and domestic animals; chemical, physical, and noise pollution; and many other stressors and pathogenic agents. The close proximity between tapirs and domestic livestock in several parts of the global distribution of the Tapirus genus creates abundant opportunities for disease transmission (Medici et al. 2014).

Over the last decades, our knowledge tapir biology and medicine has improved significantly thanks to a variety of in-situ and ex-situ research projects, observations, and scientific contributions from biologists, veterinarians, and other wildlife professionals. The line between research on captive and wild tapir biology and health is not as clear as one might

think. Understanding how tapirs live in their natural habitat improves our understanding of disease epidemiology in wild populations, but it can also provide information that helps prevent many of the health problems common in captivity. Likewise, proper management and research of captive tapirs can significantly contribute to in situ conservation programs (Mangini et al. 2012).

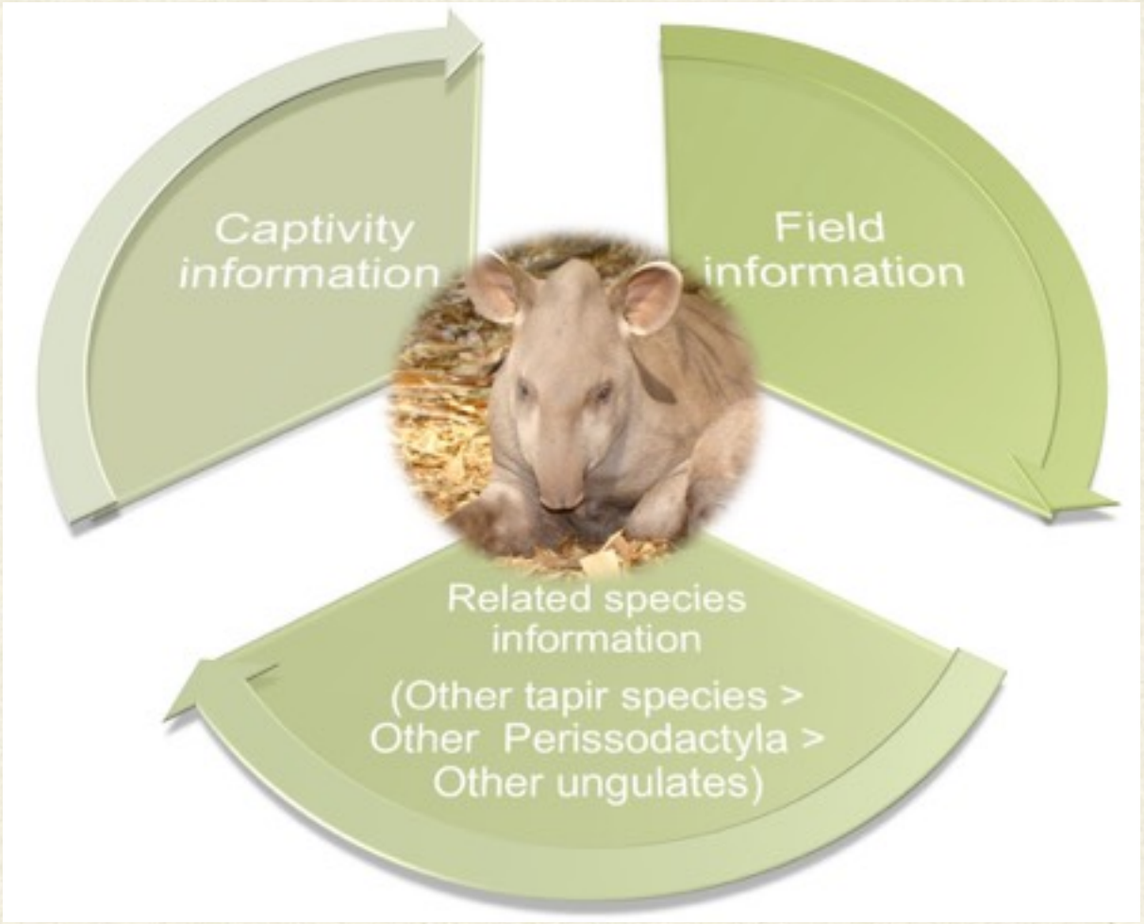

Figure 1 *- Information that should be compiled and considered for tapir health studies. Diagram: Renata Carolina Fernandes-Santos.*

The TSG is both a principle proponent of research on tapir health and an important resource for tapir health information. TSG members and colleagues frequently publish on tapir health in the TSG Tapir Conservation Newsletter and other international scientific journals, and present their work at a variety of conferences and symposiums, including the International Tapir Symposiums.

Overall, most of the information about tapir health comes from captive collections (Janssen et al. 1999; Nunes et al. 2001; Mangini et al. 2002; Janssen 2003; Mangini 2007). The physiological data reference values most commonly used for tapirs were compiled from captive tapirs (ISIS - International Species Information System, Teare 2006). In fact, there is an almost complete lack of data about health assessment in wild tapir populations. Available information from wild tapir populations comes from a long-term Baird's tapir (Tapirus bairdii) study in Corcovado National Park, Costa Rica (Hernandez-Divers et al. 2005) as well as from results of lowland tapir studies in Brazil (Furtado et al. 2010; May-Jr 2011; Medici 2010; Medici et al. 2014). Given this, the impacts of diseases on wild tapir population dynamics remains largely unknown.

Yet this is changing. The involvement of veterinarians and microbiologists in field projects is viewed as increasingly essential in many conservation programs. Indeed, tapir field projects are becoming more transdisciplinary in order to meet the variety of threats to the survival of wild populations, including those related to health. Likewise, wildlife health professionals are working to maximize the amount of information available on tapir health. Veterinarians can significantly increase the amount and improve the quality of scientific data gathered through field projects, and can make a variety of important contributions in the field:

1. Veterinarians can design adequate tapir health assessments.

2. Veterinarians are professionals with specialized knowledge and training on the capture and chemical immobilization of animals, including monitoring the well-being of anesthetized animals and dealing with possible complications from anesthesia.

3. Veterinarians are familiar with diseases affecting tapirs and other ungulates in their study region, and are able to evaluate and monitor epidemiological or endemic population health challenges, and elaborate disease control strategies.

4. Veterinarians are prepared to design appropriate protocols for the collection, handling and storage of the biological samples needed for diagnostic tests in disease and genetic research, and other scientific investigations.

5. Veterinarians are prepared to design appropriate protocols for the collection, handling and storage of the biological samples needed for diagnostic tests in disease and genetic research, and other scientific people.

6. Veterinarians are trained in anatomy and physiology, and are therefore essential in projects that will include any aspects of nutrition, reproduction and behavior.

7. Veterinarians are well positioned to train field personnel (biologists, para-biologists etc.) on wildlife capture and immobilization, sample collection/handling/storage, identification of diseases based on clinical signs, physical examinations living animals, the diagnosis of nutritional deficiencies and post-mortem examination.

8. In the case of re-introduction, translocation, or population restoring projects, only veterinarians are qualified to evaluate the health of all animals intended for release, which is necessary to avoid the introduction of novel pathogens and to protect the population intended for reintroduction.

Field veterinarians should be well-informed and up to date with the current concepts in wildlife medicine, conservation biology and ecology. Expertise in Conservation Medicine and the recent “Ecohealth” and “One Health” approaches is essential.

The term Conservation Medicine was coined by Koch (1996) and refers to a science created to address the global health crisis, which increasingly jeopardizes biodiversity, causing growing imbalances in ecosystem, human, animal and vegetal health (Aguirre et al. 2002). Conservation Medicine is a holistic approach that examines local ecosystem health issues as components of a larger, inter-dependent web of life in which actions and phenomena at certain levels ripple throughout the system. This paradigm mandates a transdisiciplinary approach capable of addressing various causes and effects at multiple levels (Aguirre et al. 2002).

The formation of transdisciplinary teams is fundamental to improve conservation projects and maximize their yield.

In response to the growing health implications of environmental degradation, Conservation Medicine includes examining the relationship among (a) changes in climate, habitat quality, and land use; (b) emergence and re-emergence of infectious agents, parasites, and environmental contaminants; and (c) maintenance of biodiversity and the

ecosystem functions that sustain the health of plant and animal communities, including humans (Aguirre et al. 2002; Aguirre et al. 2012).

To apply the Conservation Medicine paradigm, it is extremely important that disease specialists equipped to assess the health-related components of such interactions are involved in the design of tapir field projects. Experts such as veterinarians, biologists, bacteriologists, virologists, and geneticists are the best qualified professionals to identify health issues and properly design a health assessment study and sampling protocols given their knowledge of:

a) Population level health threats.

b) The types of etiologic agents that normally cause clinical disease.

c) The role diseases normally play in tapir population dynamics.

d) The local domestic animal diseases and how/if they can affect tapirs.

e) The potential for tapirs to function as reservoirs for domestic animal and zoonotic diseases.

f) Methods for predicting, preventing and/or controlling such diseases.

g) The possible role of anthropogenic impacts in tapir health.

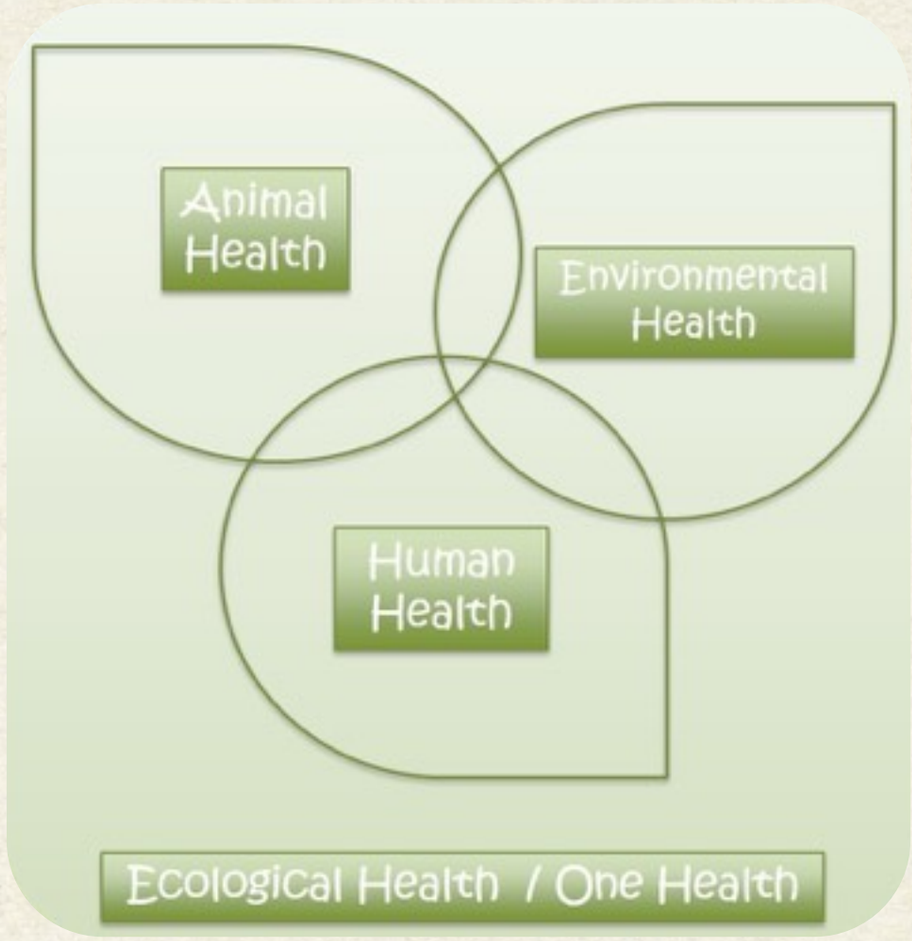

Figure 2 *- Conservation Medicine / One Health approach. Diagram: Renata Carolina Fernandes-Santos.*

Summary of Chapter:

The majority of tapir health publications report on data collected on captive tapirs. Yet this is changing as more and more tapir research and conservation initiatives espouse holistic paradigms that mandate the presence of field veterinarians. This is a positive development for the field, as both ex-situ and in-situ research are necessary to gain a complete understanding of important tapir health issues. As one component of these holistic approaches to conservation, it is essential that wildlife health researchers and professionals use an ecological approach that examines possible relationships between infectious agents, human hosts, domestic animals and wildlife, and their ecosystems. It will be important to monitor the influence of these interactions over time (Medici et al. 2014).

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Picture: Byron Jorjorian

2

Tapir Anatomy

Tapir Anatomy

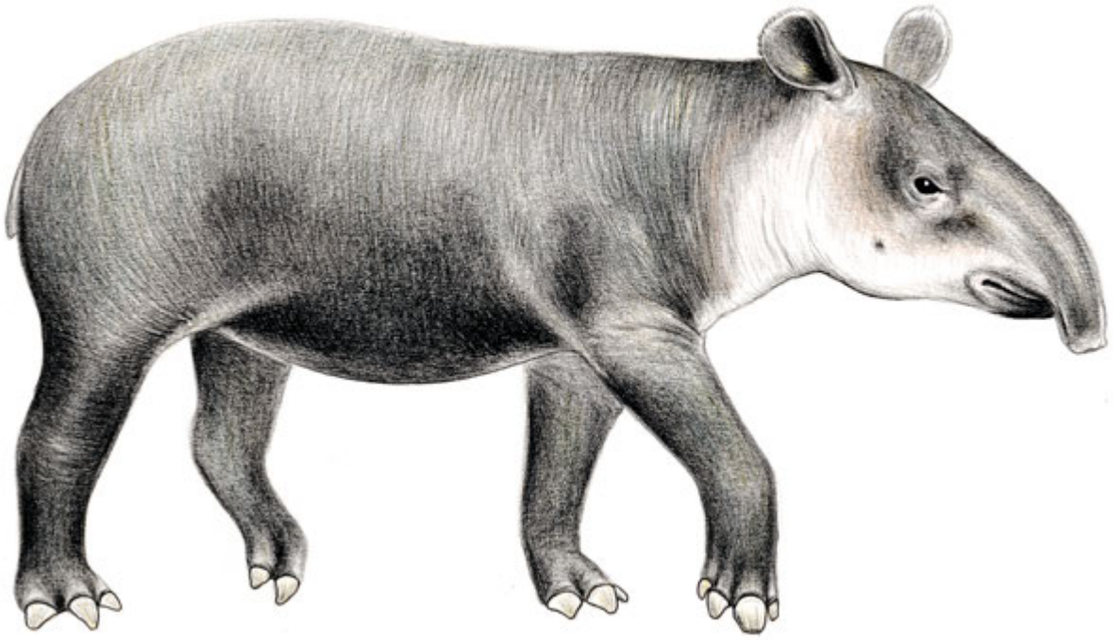
Anatomy is the branch of biology concerned with the study of the form and structure of organisms. It is most practically divided into macroscopic and microscopic anatomy. Macroscopic anatomy, or gross anatomy, is the examination of an organism's parts using unaided eyesight. Microscopic anatomy, also known as histology and cytology, involves the use of optical instruments in the study of the tissues and cells of various structures. This chapter will provide basic information about general macroscopic anatomy of tapirs, as well as important anatomical adaptations of the species.

Tapirs have robust physical structure and are quite large compared with many other mammals. In the ecosystems where they occur, they are generally one of the largest terrestrial mammals. The Malayan Tapir, Tapirus indicus, weighs between 280-400 kg and is the largest of all tapir species. The second largest tapir species is the Central American Tapir, Tapirus bairdii, which weighs between 250-350 kg. The Lowland Tapir, Tapirus terrestris, is the third largest, weighing 180-300 kg. The Mountain Tapir, Tapirus pinchaque, weighs 150-200 kg and is the smallest of the four tapir species. Please see further details in TABLES 1 and 2. Female tapirs are generally larger than males, but there is no apparent sexual dimorphism.

Gallery 1 - Tapir Species Illustration

Tapirus pinchaque

Tapirus terrestris



Tapirus bairdii

Tapirus indicus

Tapirs are solid, massive animals that are round in the back and tapered in the front, which makes them well suited for rapid movement through thick underbrush. The internal anatomy and physiology of tapirs is similar to that of the domestic horse and other Perissodactyla.

Table 1 *- Average body mass of different tapir species (Shoemakeret al. 2003 - Guidelines for Maintenance and Management for Tapirs in Captivity. IUCN/SSC Tapir Specialist Group (TSG).*

Species	Male (kg)	Female (kg)
Tapirus bairdii	180-270	227-340
Tapirus indicus	295-385	340-430
Tapirus pinchaque	136-227	160-250
Tapirus terrestris	160-250	180-295

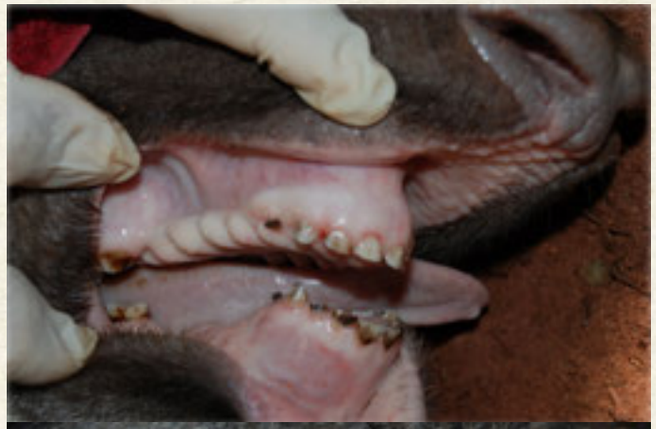
Table 2 *- Estimated body mass of wild lowland tapirs in the Atlantic Forest-AF (1996–2008) and Pantanal-PA (2008–2012), Brazil (Medici et al. 2014).*

		AF (N=44)			PA (N=68)		
Sex	Age Class	Mean (kg)	SD	N	Mean (kg)	SD	N
Female	Adult	230	32	25	221	11	20
	Sub-adult	170	7	2	190	0	8
	Juvenile	90	-	1	-	-	-
Male	Adult	205	38	11	210	18	16
	Sub-adult	189	17	4	190	0	15
	Juvenile	90	-	1	140	25	9

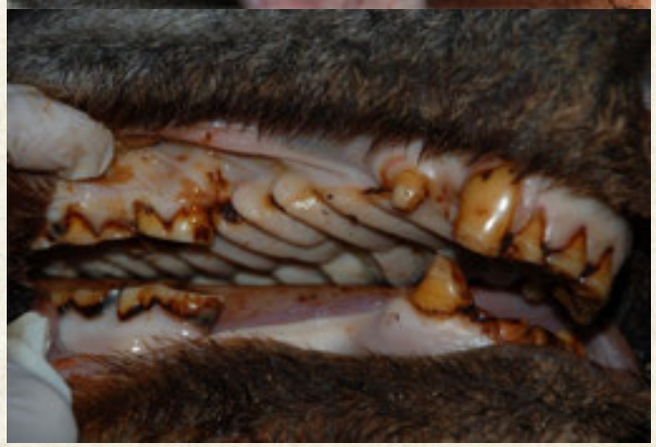
SD = standard deviation

The dental formula of adult tapirs is similar to that of equids: I 3/3, C 1/1, P 4/3, M 3/3 = 42. Males and females have similar teeth. The upper third incisors are large and well-developed, and the upper canines are reduced and separated from the incisors by a narrow diastema. The lower third incisors are reduced and the lower canine is well-developed, occluding with the canine-like upper third incisors. The incisors are chisel-shaped and the canines are conical. All cheek teeth lack cement. They are low-crowned and strongly lophodont. There is also a large diastema between canines and premolars in both jaws.

Gallery 2 - Dentition



Dentition of a juvenile lowland tapir Tapirus terrestris. Photo: Patrícia Medici.



Dentition of a fully grown adult lowland tapir Tapirus terrestris. Photo: Patrícia Medici.

Gallery 3 - Skull

Lowland tapir skull. Photos: Renata Carolina Fernandes-Santos

Lowland tapir skull. Photo: Temaiken Foundation

Tapirs have relatively long, laterally compressed skulls with a high braincase and convex profile. The nasal bones are short, arched, and freely projecting. The nasal opening is very large. Tapirs have a short, fleshy proboscis derived from muscle and soft tissues from the snout and upper lip. The proboscis is highly mobile and sensitive to touch, and is important for food manipulation and ingestion. It helps tapirs to feed on branches and leaves. Apart from its use as a gripping organ, the proboscis also comes in handy for sniffing and feeling food or other items.

The neck of tapirs is quite thick and tough. This provides the animal protection when it moves through dense, often prickly undergrowth, and possibly serves as a defense mechanism against predators, which seize the back of the neck. Lowland Tapirs have a well-developed, prominent saggital crest that runs from the base of the muzzle to the middle of the back. It is derived from fat and soft tissues and covered by long black hair. The tail is slightly prominent.

The tapir's digestive system consists of a small gut, a well developed cecum and colon, and lacks a gallbladder. The kidneys are not lobulated and, as in other water-associated ungulates, its cortex represents about 80% of the renal mass in adults.

Tapirs have pharyngeal guttural pouches similar to that of the domestic horse. The parietal and visceral pleura are normally thick and prominent, although Malayan tapirs have anatomic fibrous connective tissue between the lung and chest wall that can be mistaken for pathological adhesions. The jugular vein is found deeply in the laterals of the trachea.

The feet are mesaxonic, that is, most of the weight is borne on the middle toe. The forefoot has four main digits; the smallest one is locate behind three more prominent digits and only touches the ground when the tapir is walking on a soft substrate. The hindfeet have three digits. All of the toes are hoofed. Tapirs' splayed feet help them walk in muddy and soft ground. The weight of the body is borne by an elastic cushion under the feet and the central digits; these are the most prominent features in tapir footprints. T. pinchaque nails are comparatively longer than the other species (Mangini 2007).

Figure 3 *- A) Hindfeet (three digits); B) Forefoot (four digits); C) Tapir footprints on sand. Photos A and B: Patrícia Medici; Photo C: Renata Carolina Fernandes-Santos.*

Both thoracic and pelvic members of tapirs are extremely robust and well developed to support the body structure and weight. Bones are proportionally shorter than in other animals, and the locomotory members present a strong muscular structure. In general, tapirs have osteological and miological characteristics similar to equids; however, some morphological differences and unique anatomical adaptations are evident (Borges 2013; Pereira 2013).

The images bellow show aspects of osteology and miology of the thoracic and pelvic members of lowland tapirs in details. For further information about the anatomy of tapir's locomotory members please contact: André Luiz Quagliatto Santos (quagliatto.andre@gmail.com); Daniela Cristina Silva Borges (danybio@hotmail.com); and Saulo Gonçalves Pereira (saulobiologo@yahoo.com.br).

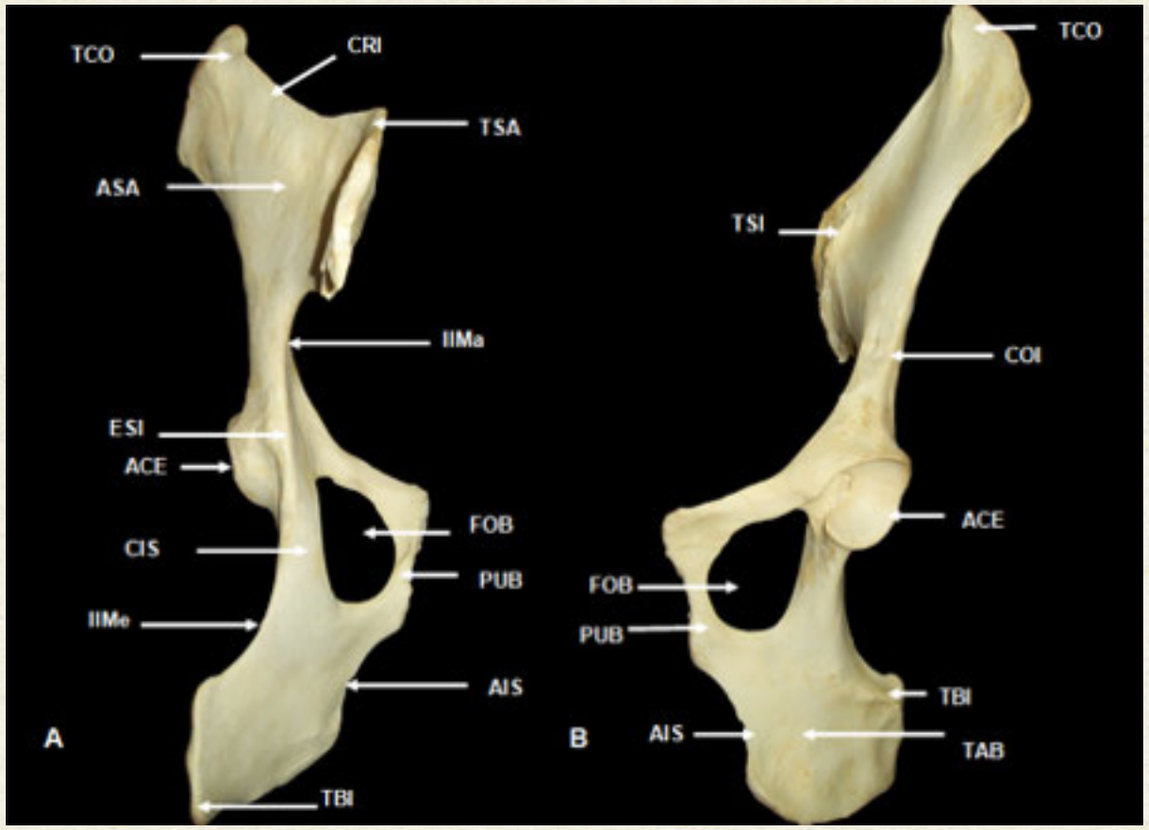


Figure 4 *– Hip bone Tapirus terrestris. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

(A), dorsal view; (B) ventral view. FOB, Foramen obturatum; ACE, Acetabulum; ASA, wing ilium; TCO, Tuberositas coxae; TBI, tuberositas isquiatic; PUB, pubis; ESI, spine isquiátic; CIS, corpus ossis ischii COI, corpus ossis ilium; TSI, Tuber sacrale iliaca; TAB, tabula; TSA, Tuber sacrale; CRI, crista aliaca; IIMa, Incisura ischiadica minor; IIMe, Incisura ischiadica maior; AIS, arco isquiatic,

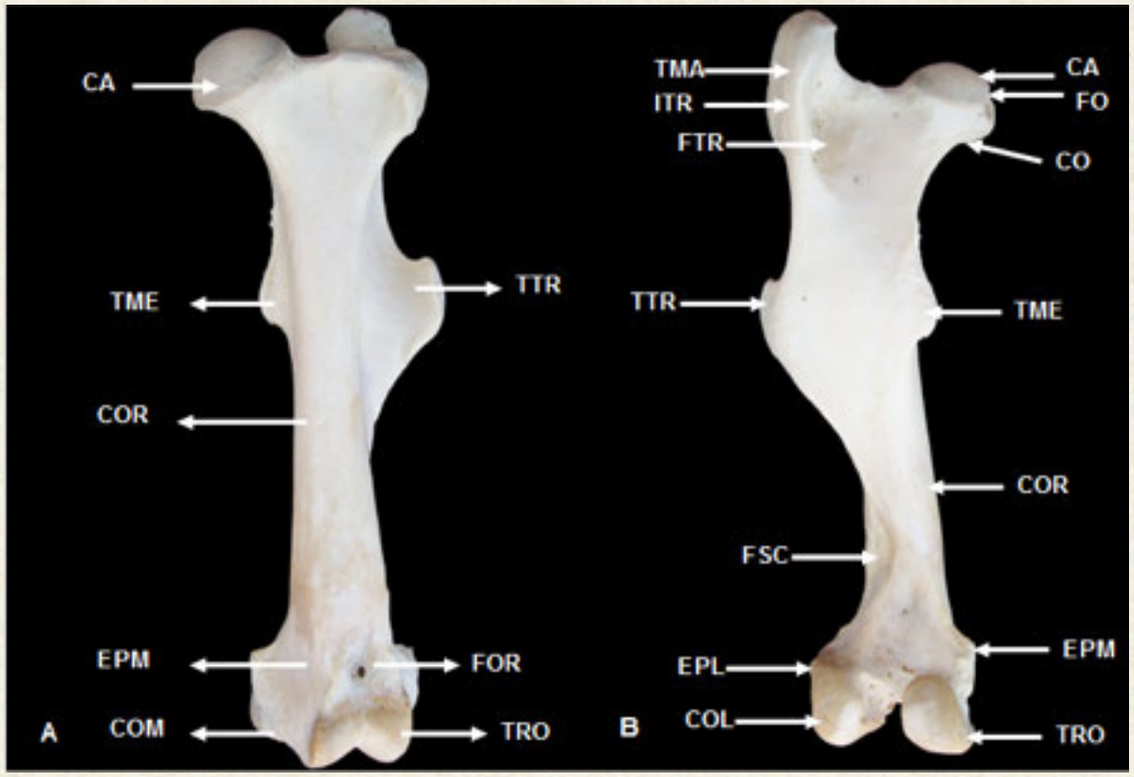


Figure 5 *– Femur of Tapirus terrestris. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

(A), cranialis view; (B), caudalis view. TRO, trochlea; FOR, foramen nutricio; COM, condylus medialis; EPM, epioôndylus medialis; TME, trochanter minor; TTR, third trochanter; CA, alla; CO, colo; TMA, trochanter major; COL, condylus lateralis; EPL, epicondylus lateralis; FSC, fossa supracondylaris; FTR, fossa trochanterilaris, FO, fovea; FOI, fossa intercondylaris, ITR, incisura trochanteralis, COR, corpus femur.

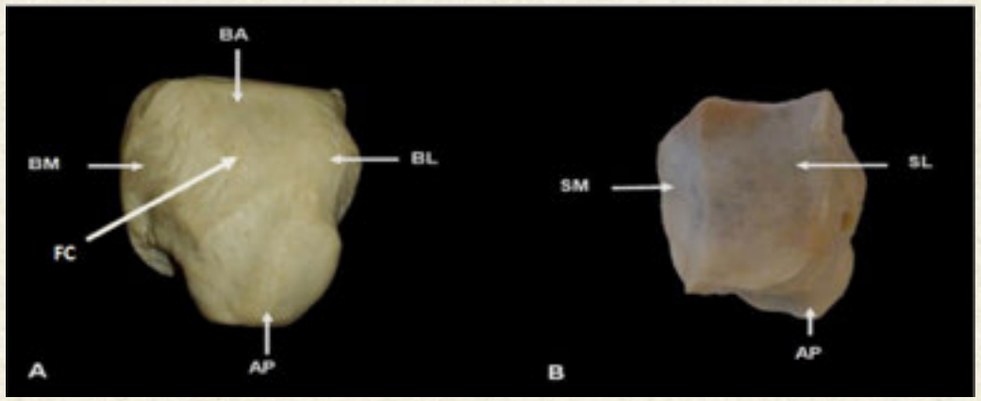


Figure 6– *Patella Tapirus terrestris. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

A), cranialis view; (B), caudalis view. BA, basis; BM, Margo medialis; AP, apex; BL, margo lateralis; SM, surface medialis facies articularum the patella; SL, surface lateralis da facie articularum the patella; FC, facie cranialis.

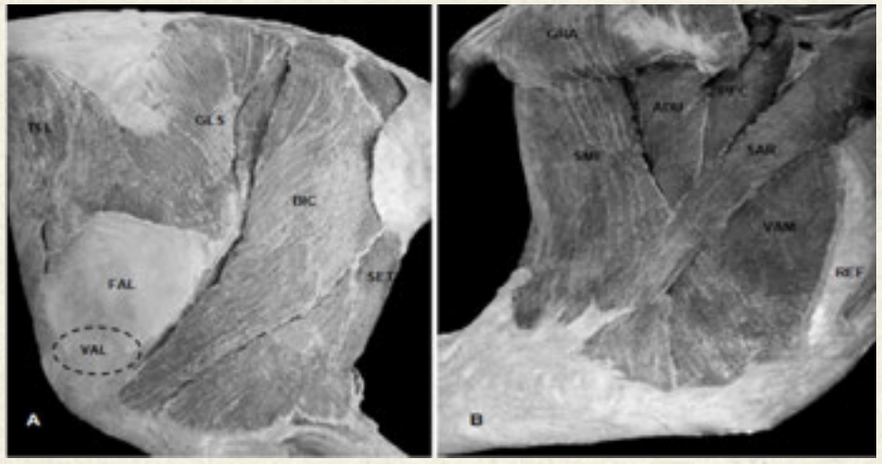


Figure 7– *Coxae muscles of Tapirus terrestris. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

(A) Surface side view; (B) superficial medial view.TFL, m. tensor fasciae lata; BIC, m. biceps femorales; SET, m. semitendinosus; FAL, fasciae lata; GLS, m. gluteus [glutaeus] superficialis, SME, m. semimembranosus; ADU, m. adductor; SAR, m. sartorius; PEC, m. pectineus; VAM, m. vastus medialis; REF, m. reto femoralis; GRA, m. gracilis; VAL, m. vastus lateralis.

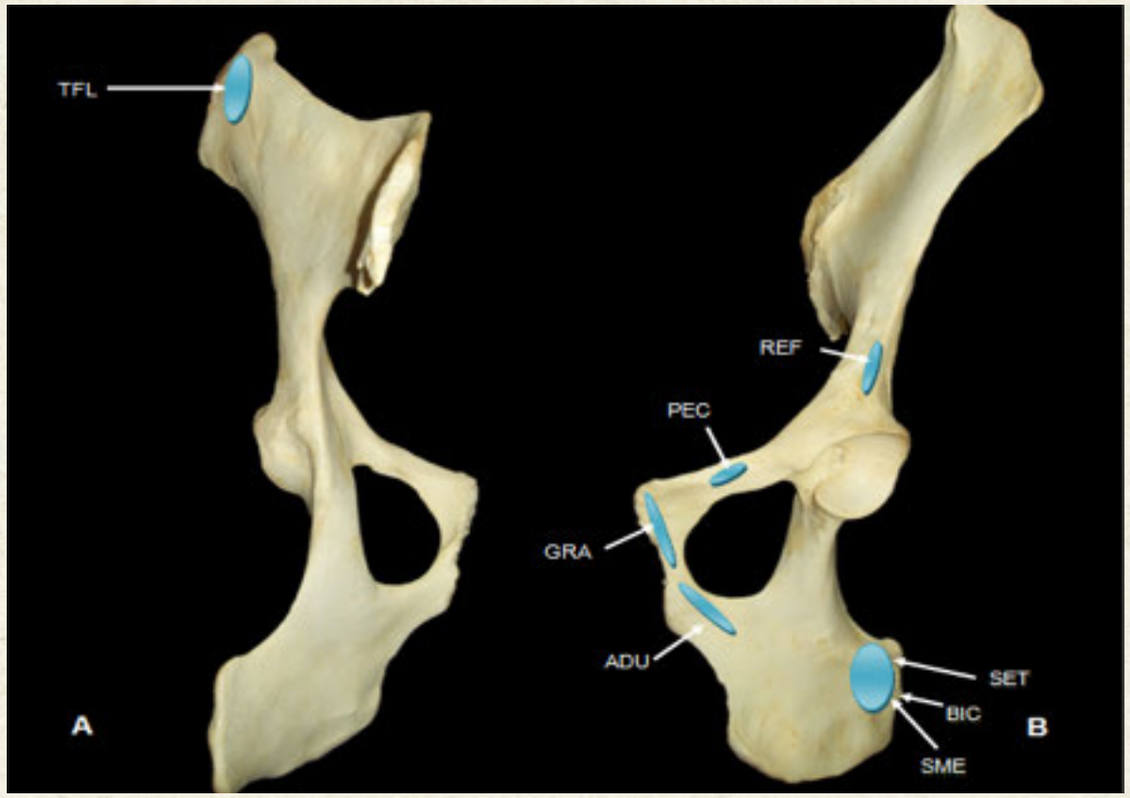


Figure 8*- Hip bone Tapirus terrestris. Origins of the coxae muscles. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

(A), dorsalis view; (B), ventralis view SME, m. semimembranosus; SET, m. semitendinosus; GRA, m. gracilis; ADU, m. adductor; PEC, m. pectineus; REF, m. reto femoris; TFL, m. tensor fasciae lata; BIC, m. biceps femoris.

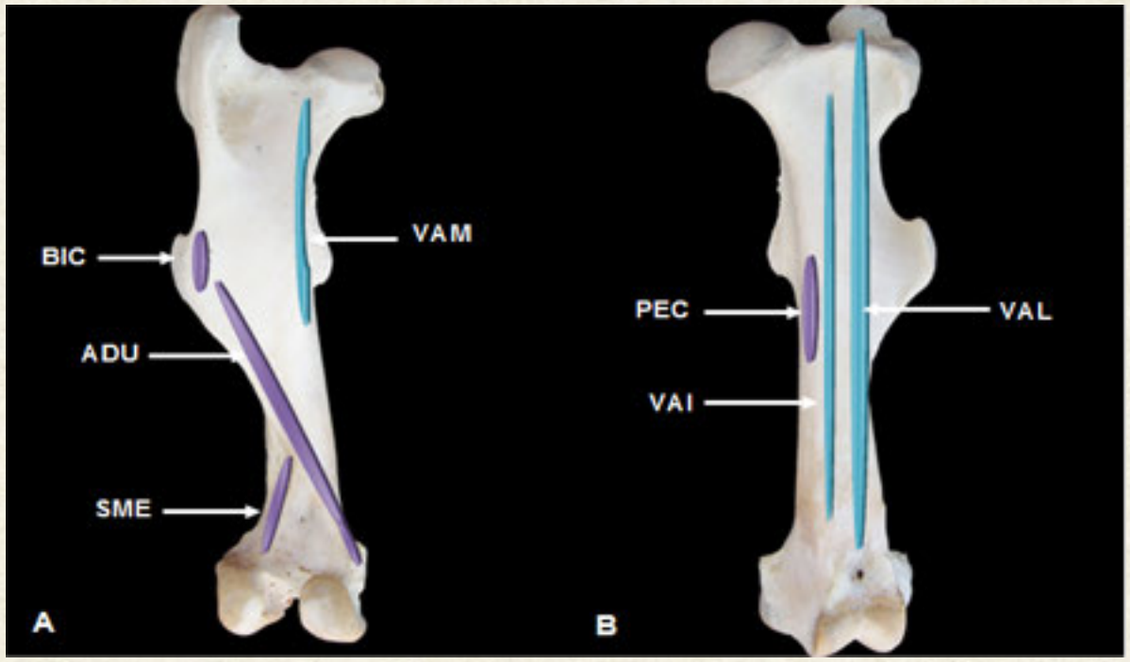


Figure 9 *– Femur Tapirus terrestris. Origins (blue) and insertions (purple) of the coxae muscles. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

(A), caudales view; (B), cranialis view. SME, m. semimembranosus; ADU, m. adductor; BIC, m. biceps; VAM, m. vastus medialis; PEC, m. pectineus; VIN, m. vastus intermedialis; VAL, m. vastus lateralis.

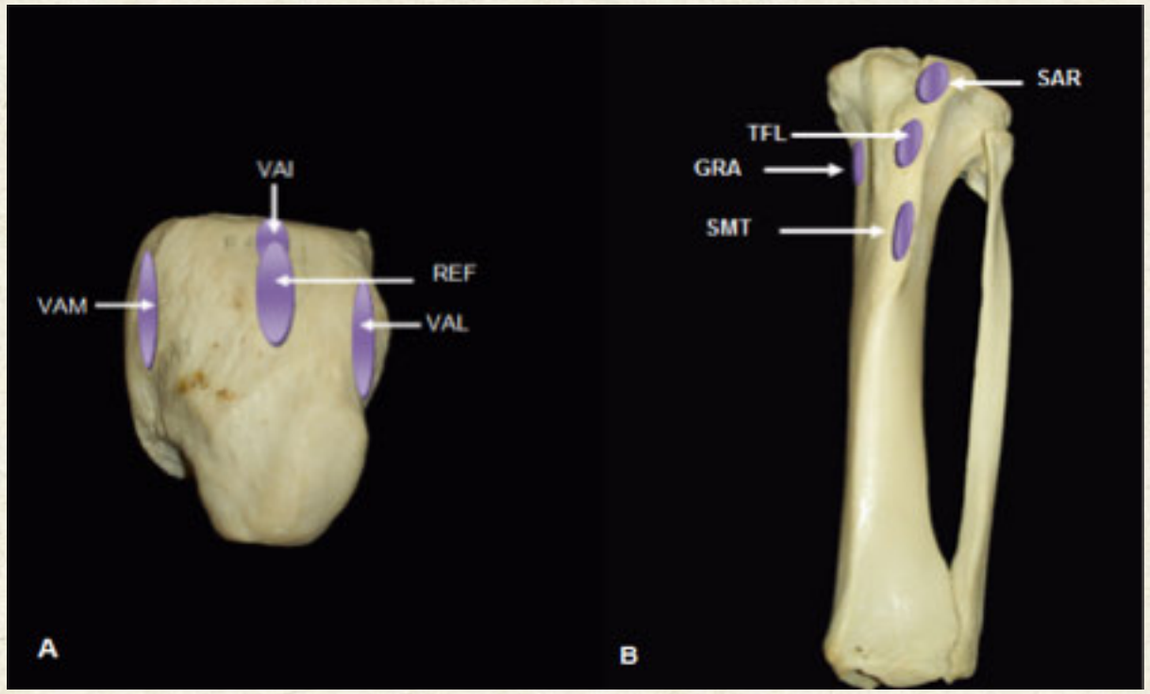


Figure 10 *- Patella, tibia and fibula Tapirus terrestris. Insertions of the muscles of the coxae.. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

A), cranialis patela view; (B), cranialis tibia e fibulae view VAM, m. vastus medialis; VAI, m. vasus intermadialis; REF, m. reto femoralis; VAL, m. vastus lateralis; SAR, m. sartorius; TFL, m. tensor da fasciae lata; GRA, m. gracilis; SMT, m. semitendinosus.

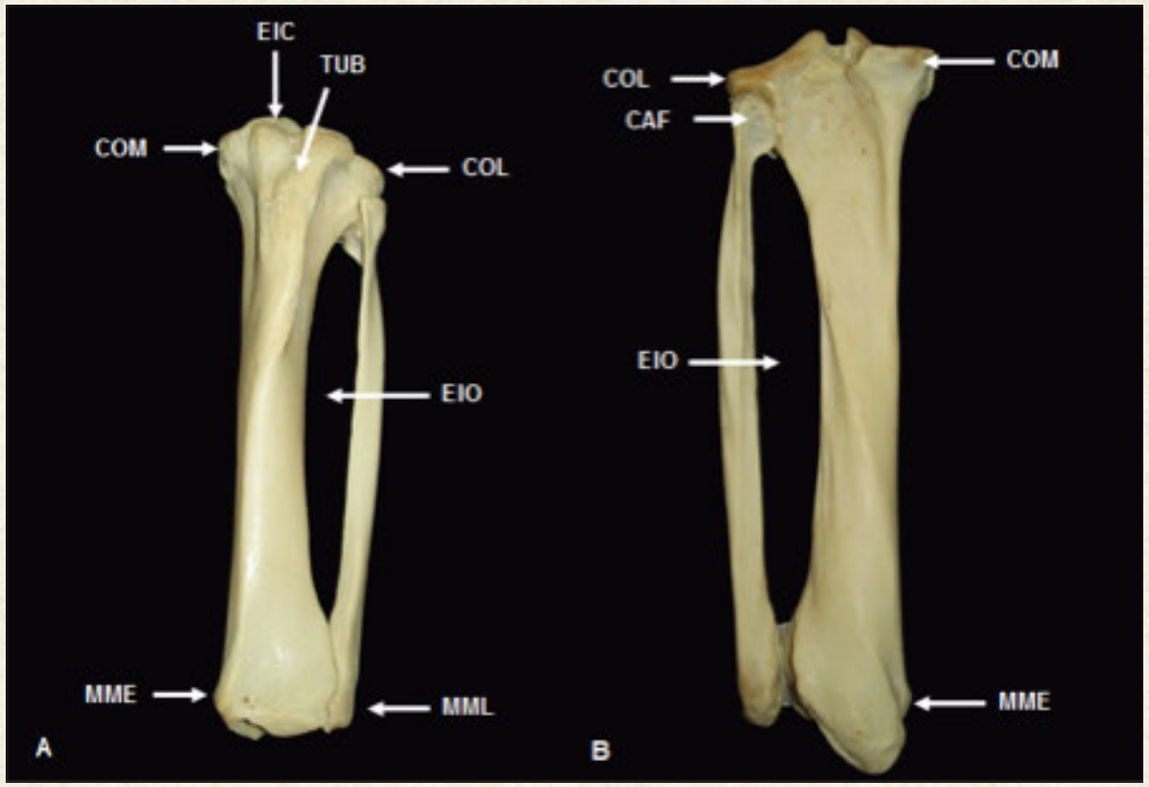


Figure 11 *– Tbia and fibula bones of Tapirus Terrestris. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

(A), cranialis view; (B), caudalis view. TUB, tuber tibia; COL, Condylus lateralis; COM, Condylus medialis; EIC, Eminentia intercondylares; MME, malleolare mediales; MML, malleolare laterales; EIO, interosseous space; CAF, caput fibulae

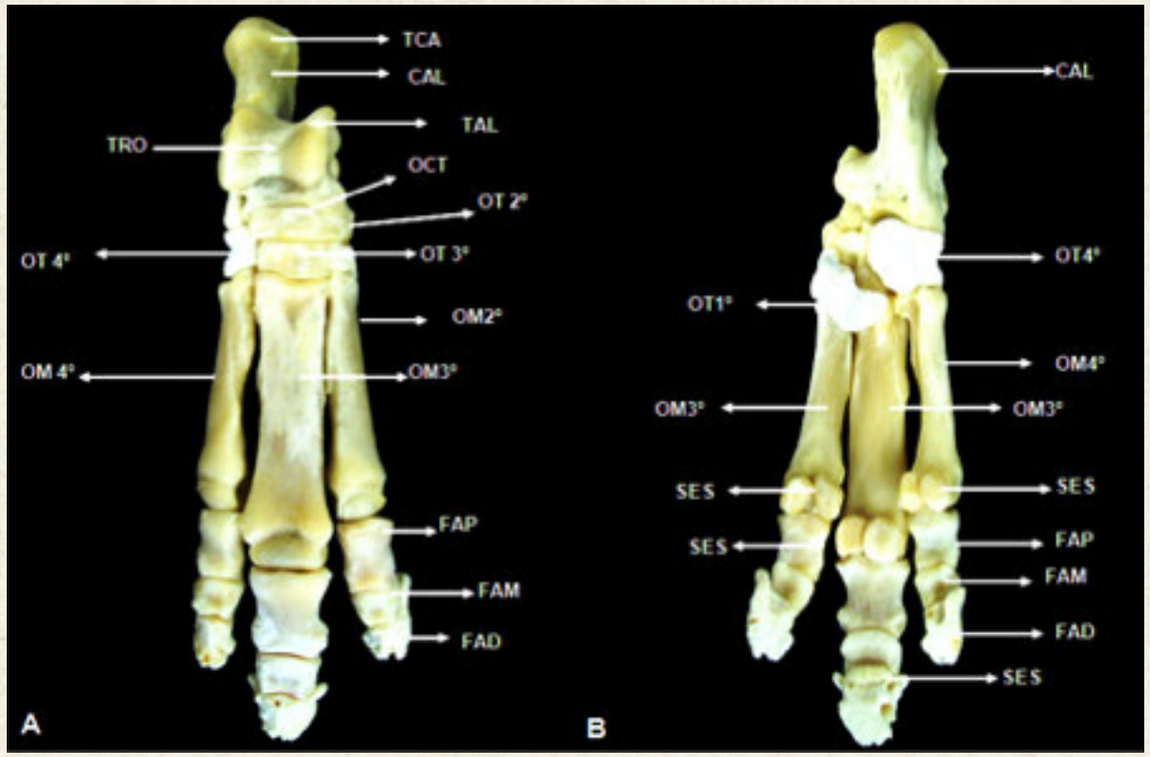


Figure 12 *– Foot bones of Tapirus Terrestris. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

(A), dorsales view; (B), pedir view. TAL, talus; CAL, Calcaneus; OCT, ossa tarsi centrale; OT1º, ossa tarsale I; OT2º, ossa tarsale II; OT3º, ossa tarsale III; OT4º, ossa tarsale IV; OM2º, ossa metatarsale II; OM3º, ossa metatarsale III; OM4º, ossa metatarsale IV; FAP, phalanx proximael; FAM, phalanx mediale; FAD, phalanx distale; TCA, tuber calcaneaum; SES, ossa sesamoideum ; TRO, tróclea,

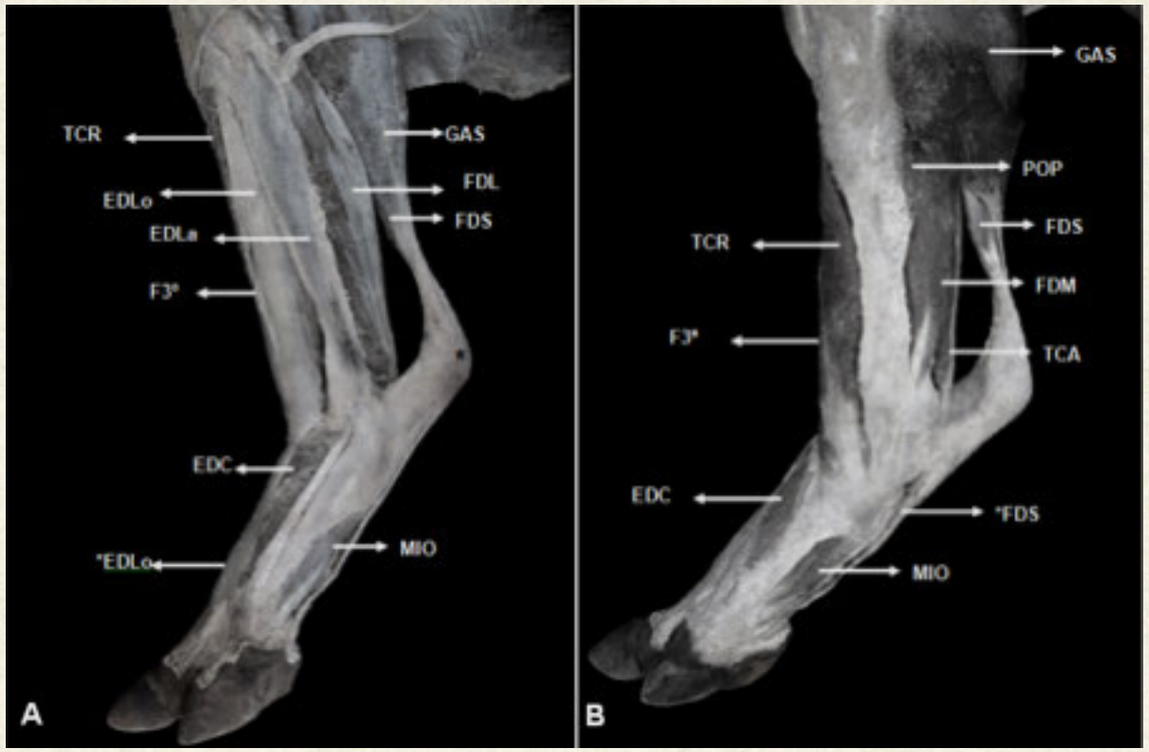


Figure 13 *– Muscles of the leg and foot Tapirus Terrestris. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

*(A), lateralis view; (B), medialis view. GAS, m. gastrocnemius; FDL, m. flexor digitorum [digitalis] lateralis; EDLa, m. extensor digitorum [digitalis] lateralis; EDLo, m. extensor digitorum [digitalis] longus; F3º, m. fibularis 3º EDC, m. extensor digitorum [digitalis] brevis; FDS, m. flexor digitorum [digitalis] superficialis; TCR, m. tibialis cranialis; *EDLo, tendo. extensorum digitorum longus; *FDS, calcaneal coverage of the superficial digiturum flexor muscle; FDM, m. flexor digitorum medialis; TCA, m. tibialis caudalis; POP, m. popliteus; MIO, mm. interossei; *, tendo the superficial digitourun flexor muscle.*

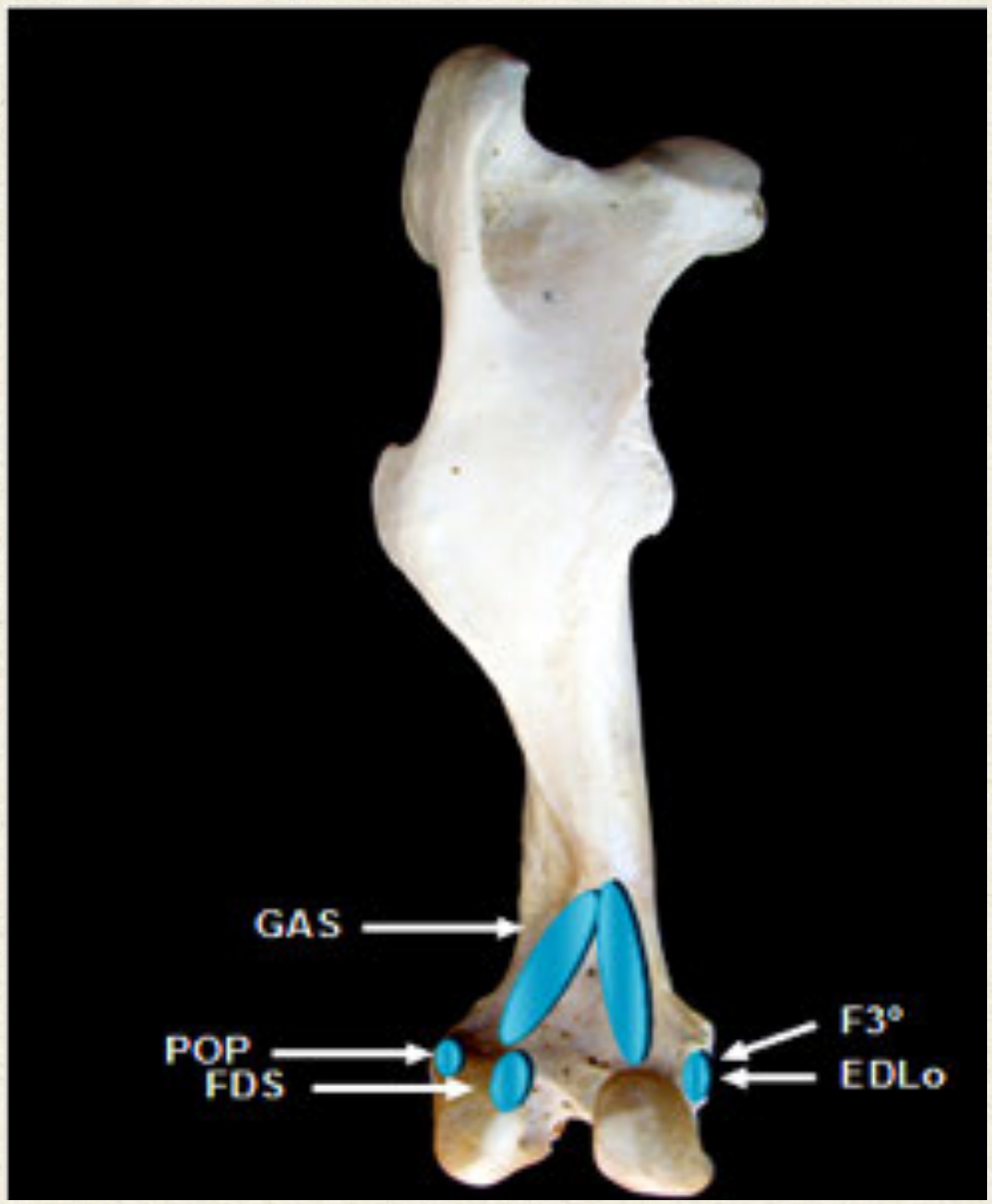


Figure 14 *– Femur bone T. terrestris, vista caudal. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

Origin of the muscles of the leg and foot. POP, m. popliteus; GAS, m. gastrocnemius; FDS, m. flexor digitorum [digitalis] superficialis; EDLo, m. extensor digiti longus; F3º, m. fibularis 3º.

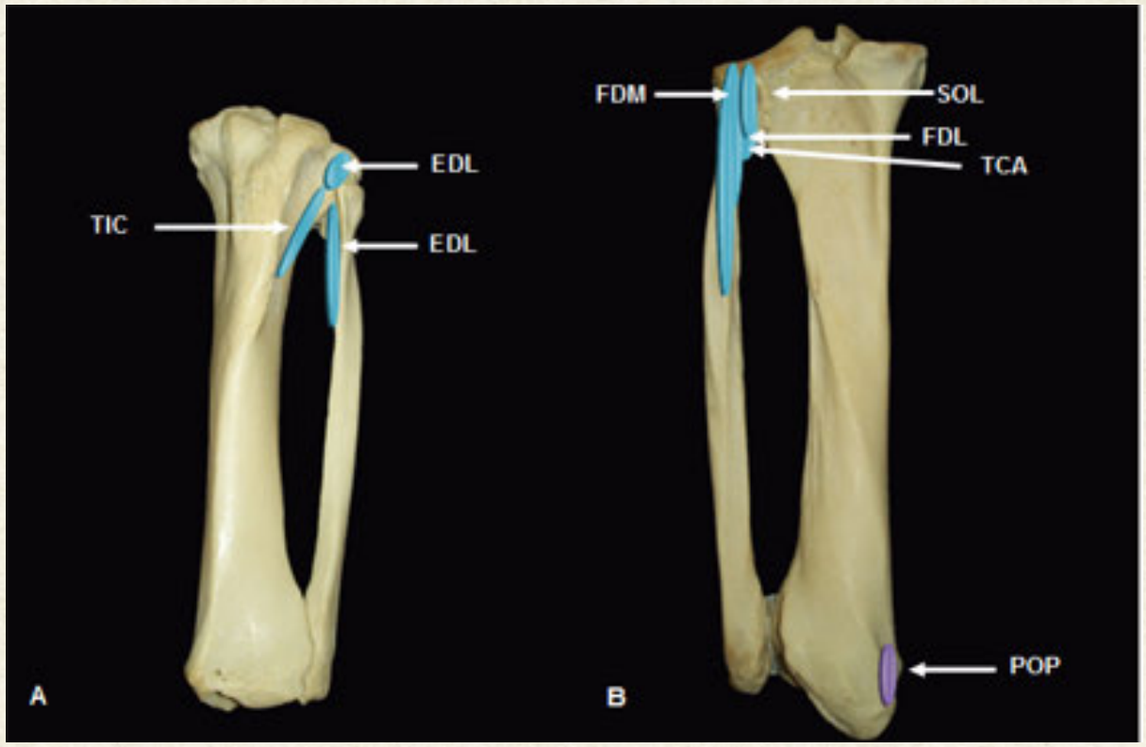


Figure 15 *– Tibia e fibulae of Tapirus Terrestris. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

Origins (blue) and insertion (purple) of the muscles of the leg and foot. (A) a cranial view; (B) Flow view.- TIC, m. tibialis cranialis; EDL, m. extensor digiti [hallucis] lateralis; FDL, m. flexor digiti [hallucis] medialis; TCA, m. tibialis caudalis; POP, m. popliteus; SOL, m. soleus; FDM, m. flexor digiti [hallucis] lateralis;.

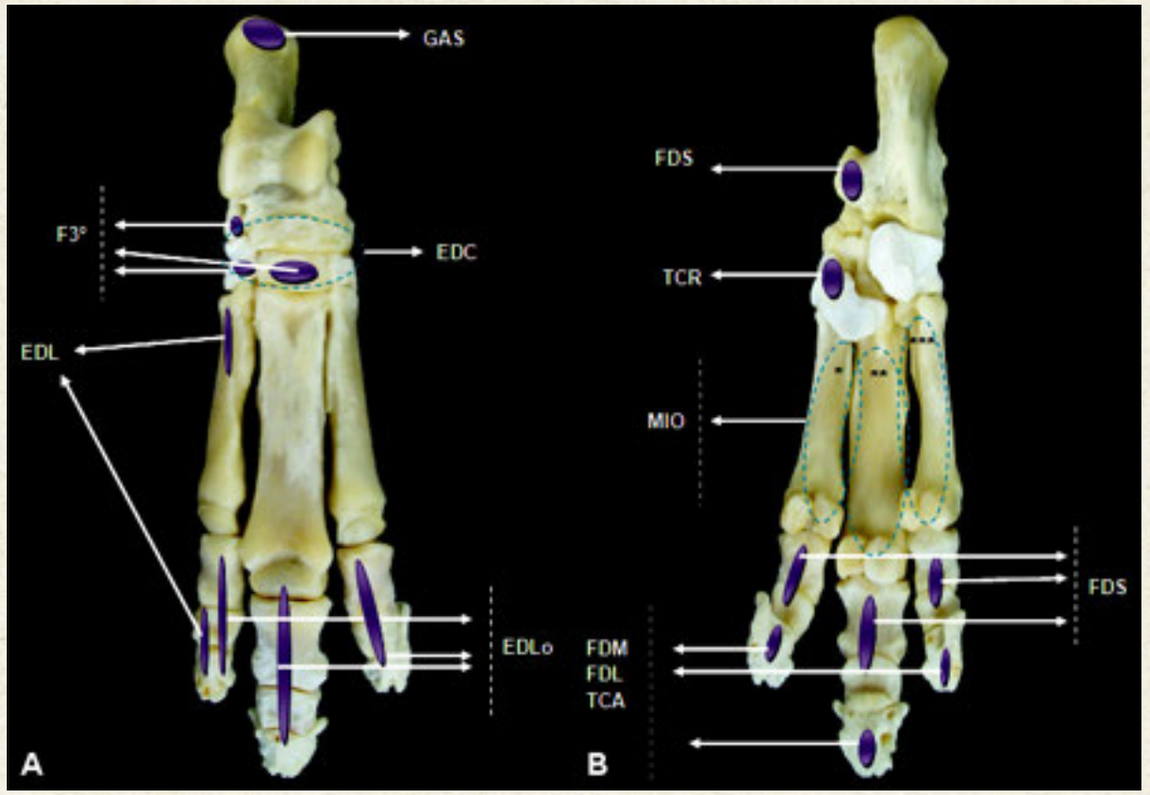


Figure 16 *– Foot bones of Tapirus Terrestris. Insertions of the muscles of the leg and foot. (Fonte: BORGES; PEREIRA, SANTOS, 2013 - LAPAS).*

*(A), dorsal view; (B) plant view. GAS, m. gastrocnemius; F3º, m. fibulares 3º; EDL, m. extensor digiti [hallucis] lateralis; EDLo, m. extensor digiti [hallucis] longus; EDC, m. extensor digiti [hallucis] lbrevis; FDS, m. flexor digiti [hallucis] superficialis; TCA, m. tibialis caudalis; FDM, m. flexor digiti medialis; FDL, m. flexor digiti lateralis; TIC, m. tibialis cranialis ; MIO, mm. interossei, * medialis, ** intermédiuns, *** lateralis.*

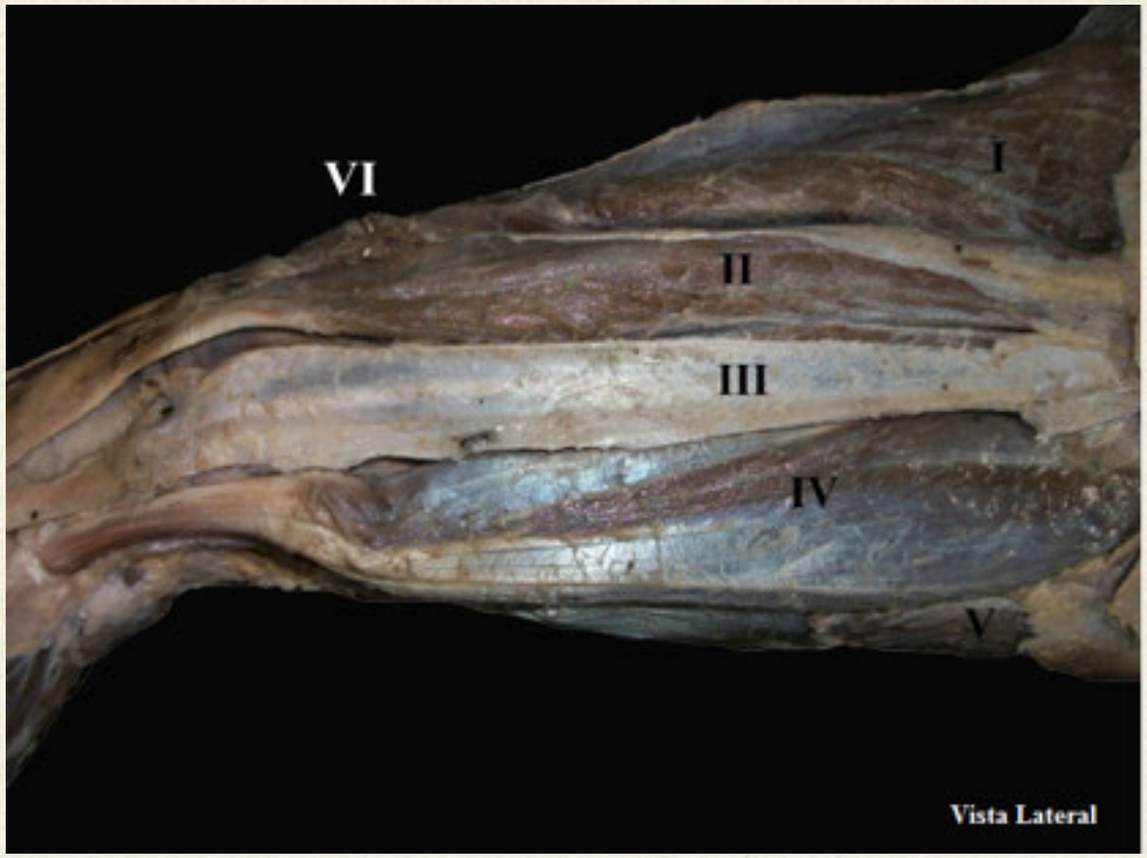


Figure 17 *– Forearm musculature of T. terrestris , side view. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legenda: (I) Extensor carpi radialis; (II) Extensor digitalis comum; (III) Extensor digitalis lateralis; (IV) Ulnaris lateralis; (V) Flexor digitalis superficialis;(VI) extensoris carpi obliqui.

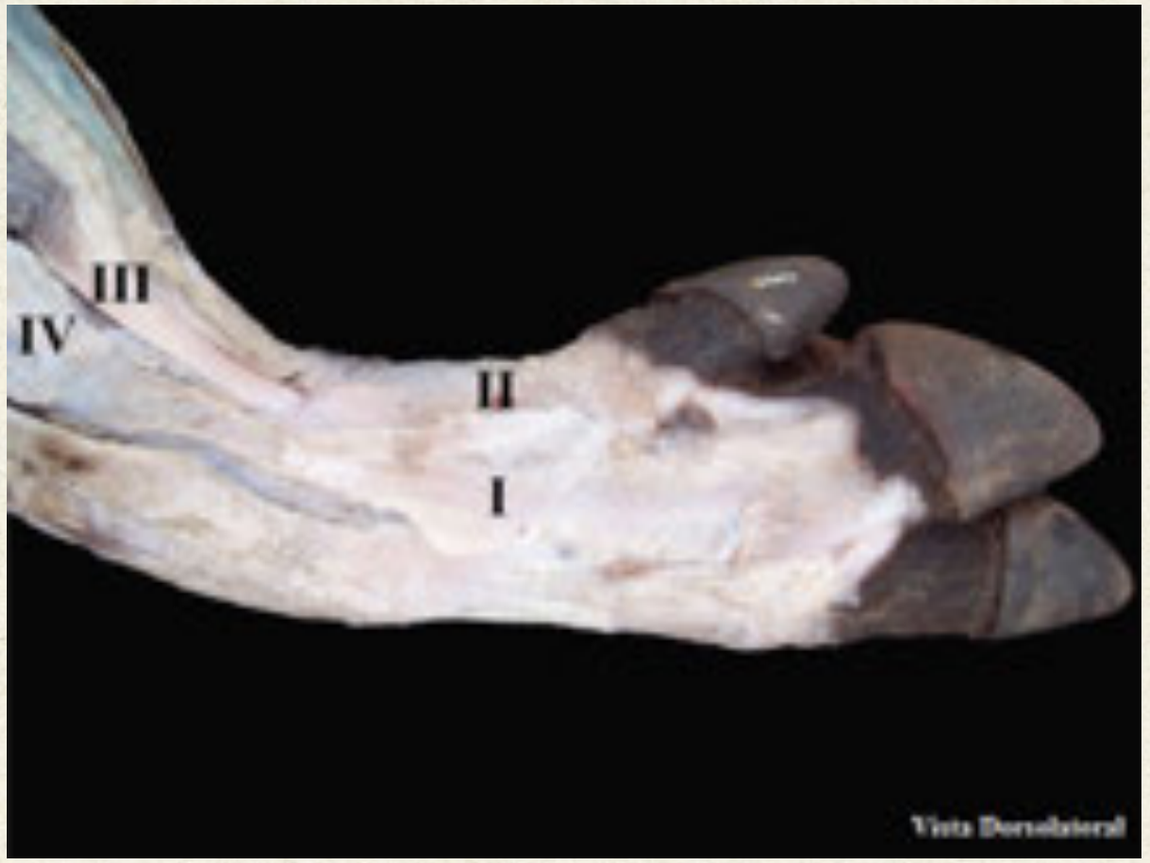


Figure 18 *– Hand muscles of T. terrestris , dorsolateral view. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legenda: (I) M extensor digitorum long the digiti II e III; (II) Extensor digitorum comum the digiti IV e V; (III) Tendo incertion the m. ulnaris Lateralis; (IV) Fascia insertion the m. digitalis lateralis.

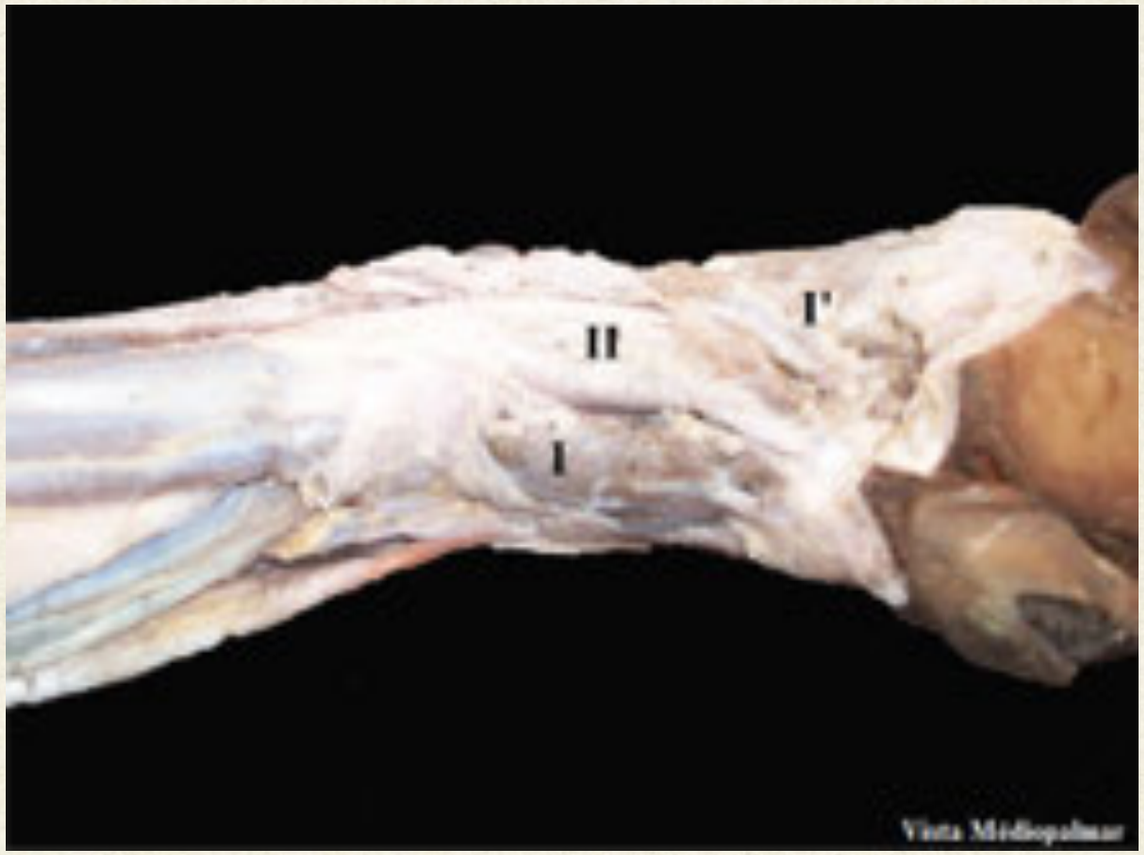


Figure 19 *– Hand muscles of T. terrestris medialis palmar view. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legenda: (I) Musculi interossei; (I') Musculi lumbricales; (II) Tendo digitalis comum.

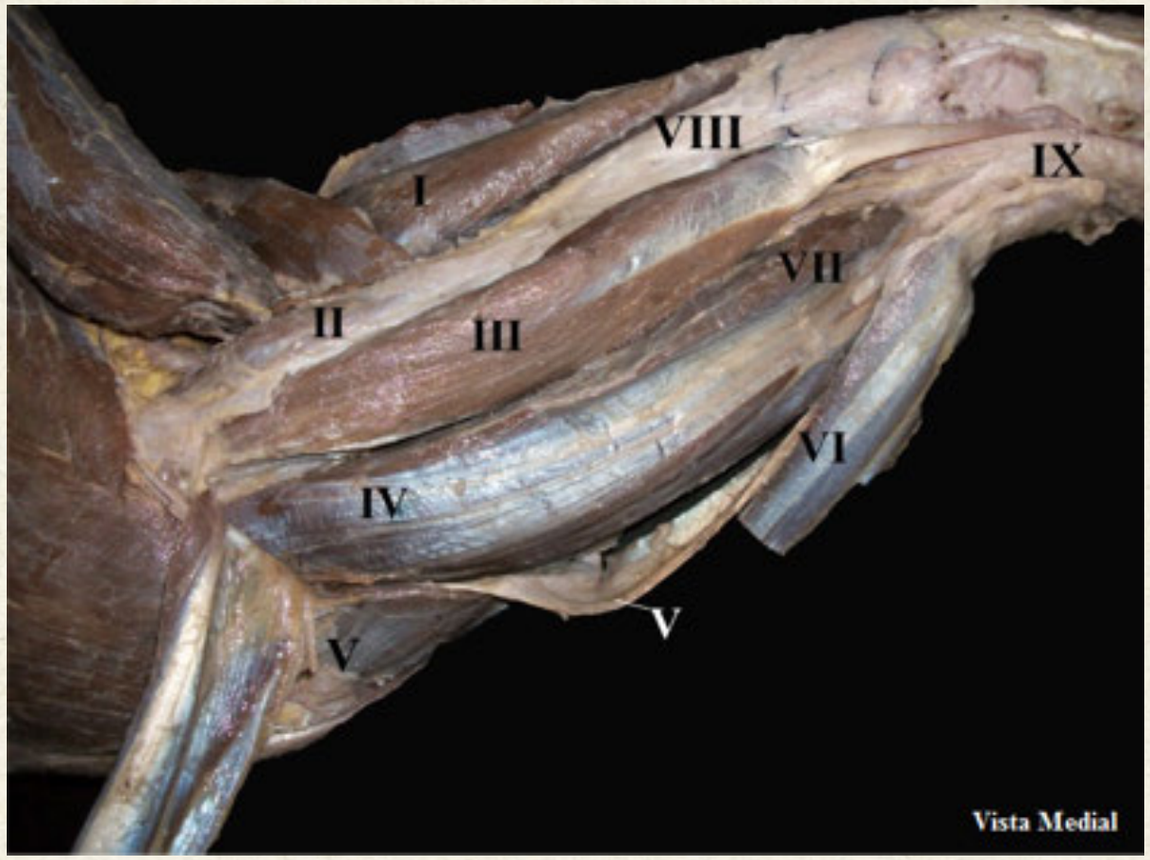


Figure 20 *– Muscles of the forearm T. terrestris, medial view. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: (I) M. extensor carpi radialis; (II) Fibrous lacertus collateral ligament; (III) flexor carpi radialis; (IV) flexor digiti profundus; (V) flexor digiti brevis (two heads); (VI) flexor carpi ulnaris; (VII) flexor digiti prfundus heads radiallis; (VIII) radius; (IX) Common tendon of the fingers.

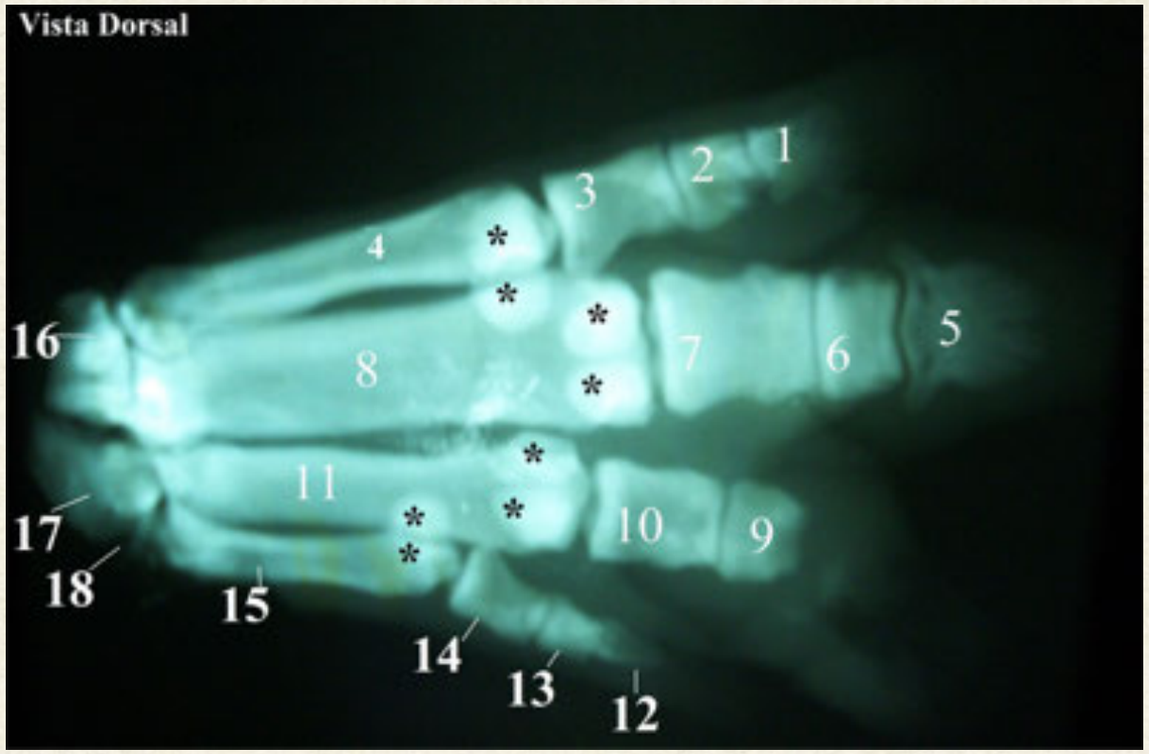


Figure 21 *- Radiological image of the hand of T. terrestris, dorsal view . (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

*Legenda: (1) Phalangis distalis the finger II; (2) Phalangis médialis the finger II; (3) Falange proximal do dedo II; (4) Metacarpo II; (5) Falange distal do dedo III; (6) Falange média do dedo III; (7) Falange proximal do dedo III; (8) Metacarpo III; (9) Falange média do dedo IV; (10) Falange proximal do dedo IV; (11) Metacarpo IV; (12) Falange dista do dedo V; (13) Falange média do dedo V; (14) Falange proximal do dedo V; (15) Metacarpo V; (16) Cárpico II; (17) Cárpico III; (18) Cárpico IV; * Sesamóides.*

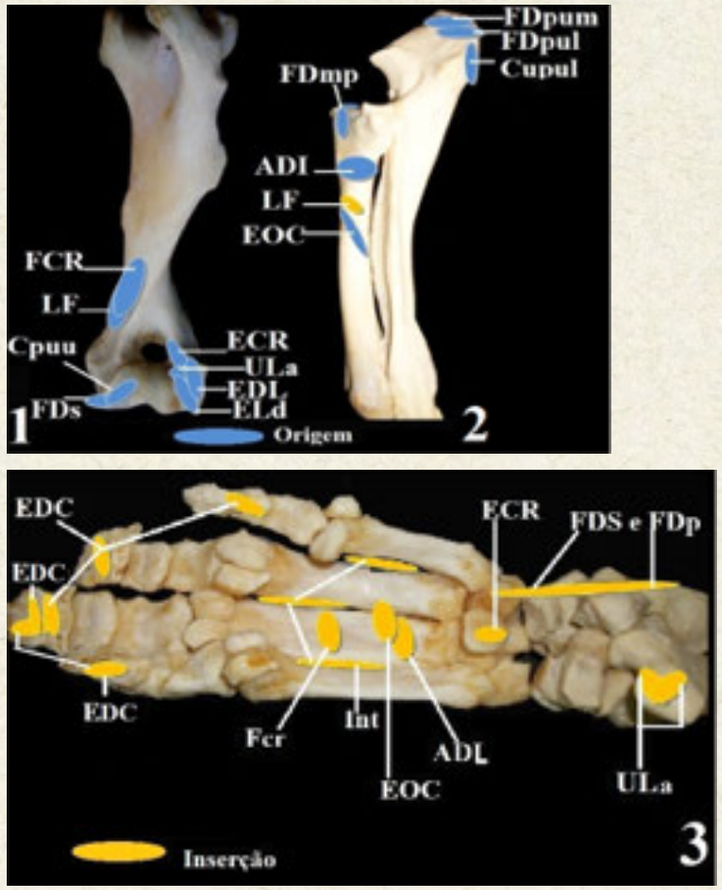


Figure 22 *– Attachment points of the forearm and hand muscles Tapirus terrestris . Yellow: point of muscle insertion . Blue : point of muscular origin (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: 1: Humero, cranialis view – (FCR) m. flexor carpi radialis; (LF) Lacerto fibrous collateral ligament of the humerus; (Cpuu) m. flexor carpo ulnares caput ulnari; (FDs) m. flexor digitii superficiali; (ECR) m. extensor carpi radialis; (ULa) m. ulnari lateralis; (EDL) m. extensor digitalis lateralis; (ELd) m. extensor digitorum longus IV e V; 2: Radius e ulna, cranial view – (FDpum) m. flexor digitalis profundo caput umeralis; (FDpul) m. flexor digitalis profundo caput ulnares; (Cupul) m. flexor carpi ulnares caput lateralis; (FDmp) m. flexor digitalis profundo; (ADL) m. long abductor; (LF) Lacerto fibrous collateral ligament of the humerus; (EOC) m. extensor oblíquum carpi; 3: Hand , palmar view – (EDC) m. extensor digitalis comum; (Int) m. interossius; (Cpu) m. carpi ulnares; (FDs) m. flexor digitalis superficialis; (FDp) m. flexor digitalis profundo; (FCR) m. flexor carpi radialis; (ADL) m. abductor levator; (ECR) m. extensor carpi radialis.

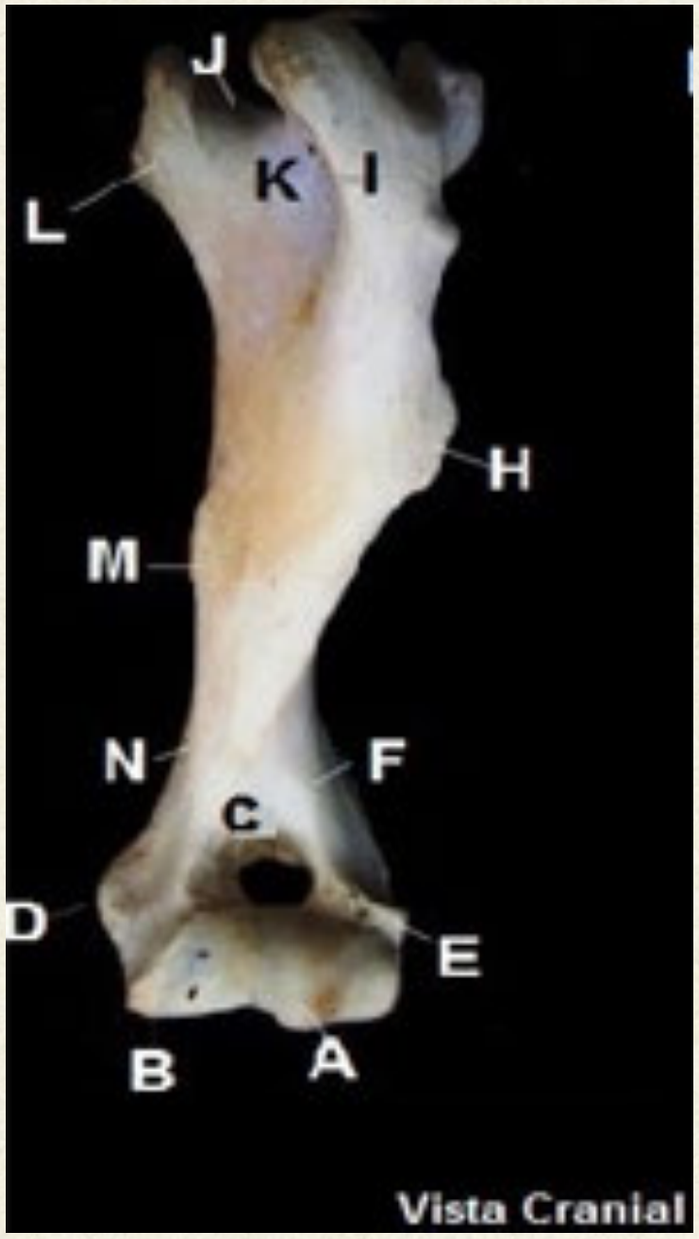


Figure 23 *– Humerus , cranial face . (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS)*

Legenda: (A) caput; (B) trochlea; (C) fossa radialis; (D) epicondylus medialis; (E) epicondylus lateralis; (F) crista epicondylus lateralis; (G) sulcus m braquii; (H) tuber deltoideus; (I) tuber major; (J) sulcus intertuberalis; (K) foramen nutricio proximales; (L) tuber minor; (M) tubero redonda major; (N) foramen nutricio distale.

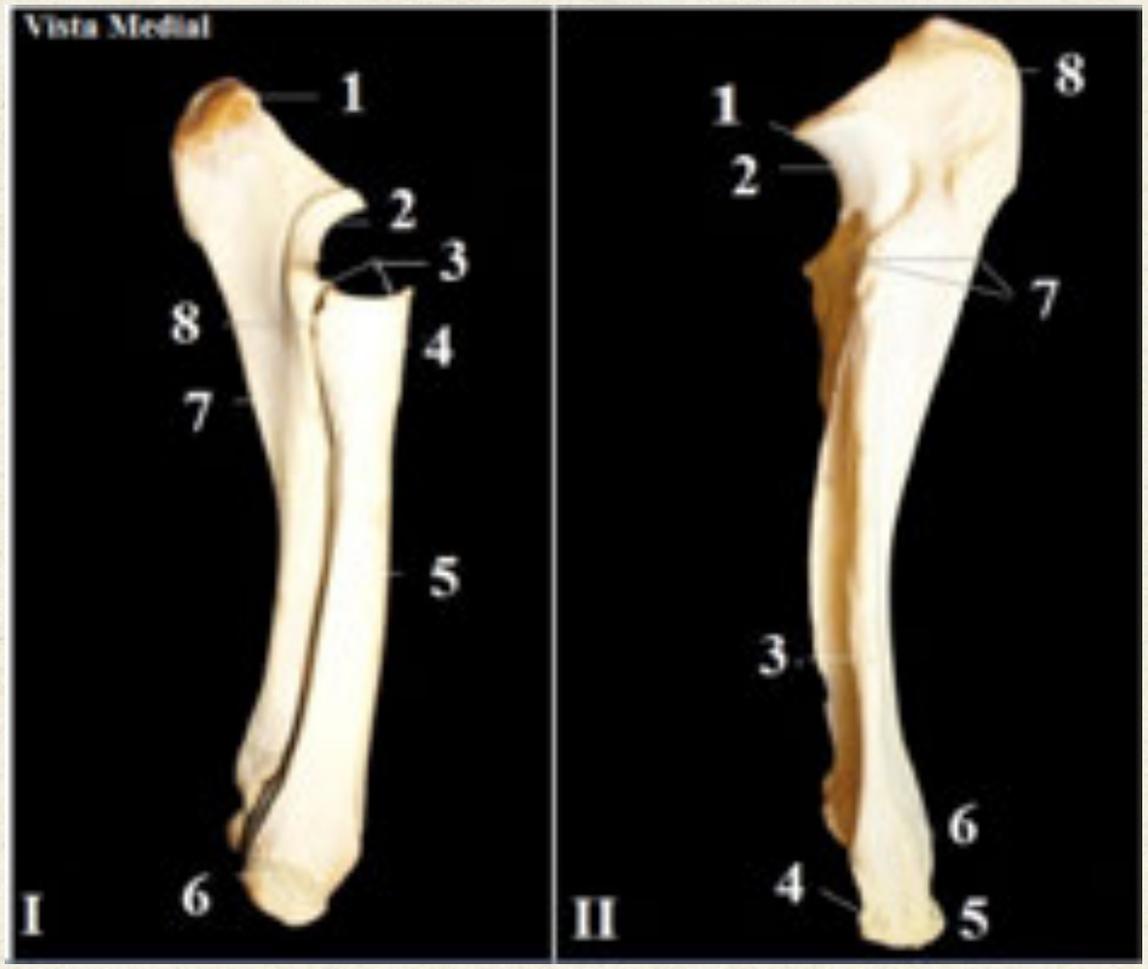


Figure 24 *– Radius and ulna bones of T. terrestris medial view. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: I: Ulna and radius – (1) Tuber olecrani; (2) Incisura trochlearis; (3) Fovea capitis radii; (4) Tuber rádius; (5) Corpus radii; (6) Carpii articular surface; (7) Corpus ulna; (8) interosseous space. II: Ulna – (1) Processus anconeus [anconaeus]; (2) Incisura trochlearis; (3) Crista ulna; (4) Forama; (5) Crista transversalis; (6) Distal end of the ulna; (7) interosseous space; (8) Tuber olecrani.

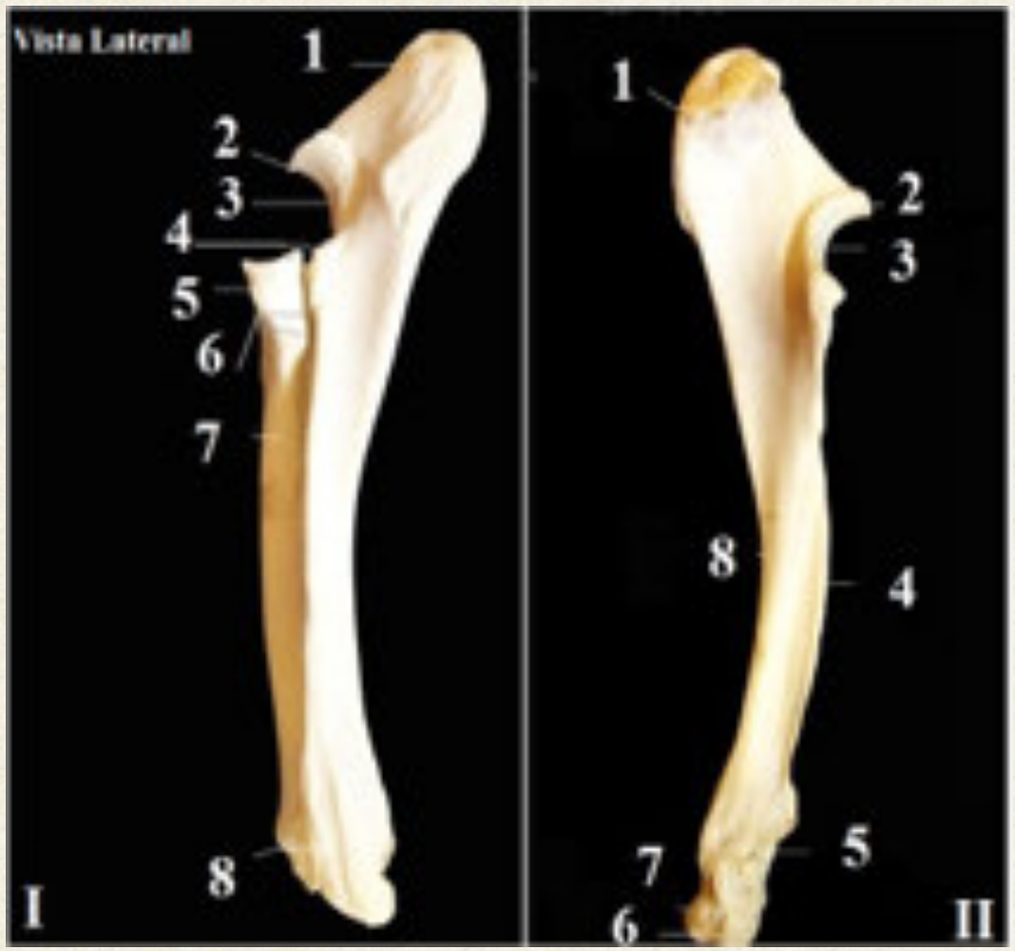


Figure 25 *– Radius and ulna bones of T. terrestris , side view ((Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: I: Radius and ulna – (1) Tuberosidade do olécrano; (2) Processus anconeus [anconaeus]; (3) Fissura trochlea; (4) interosseous space; (5) Tuber radius; (6) interosseous space; (7) Corpus do radii; (8) Sulcus tendii extensorius digitalis comum. II: Ulna – (1) Tuber olecrani; (2) Processus anconeus [anconaeus]; (3) Incisura trochlearis; (4) Crista corpus of ulna; (5) Distal end of the ulna; (6) Carpii articular surface; (7) Crista transverslis; (8) Sulcus tendii extensorius digitalis comum.

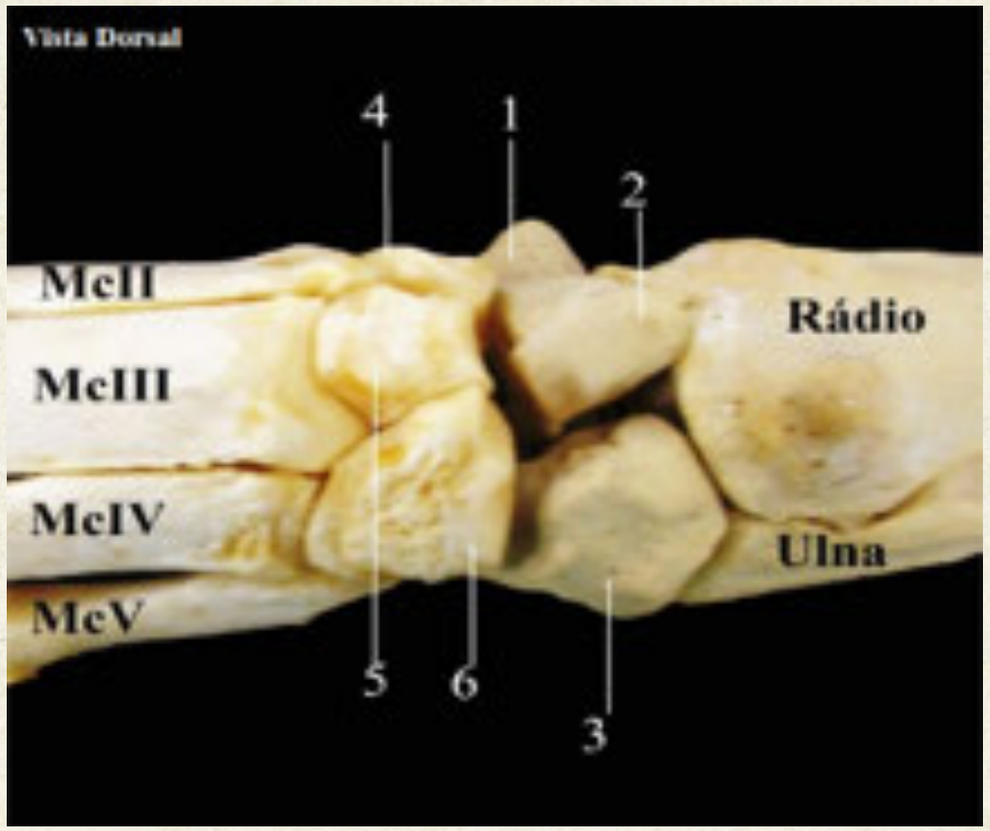


Figure 26 *– Carpal bones and metacarpal of T. terrestris , dorsal view. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: (1) Carpi radiale; (2) Carpi intermedium; (3) Carpi ulnare; (4) Carpale II; (5) Carpale III; (6) Carpale IV; (McII) Metacarpale II; (McIII) Metacarpale III; (McIV) Metacarpale IV; (McV) Metacarpale V.

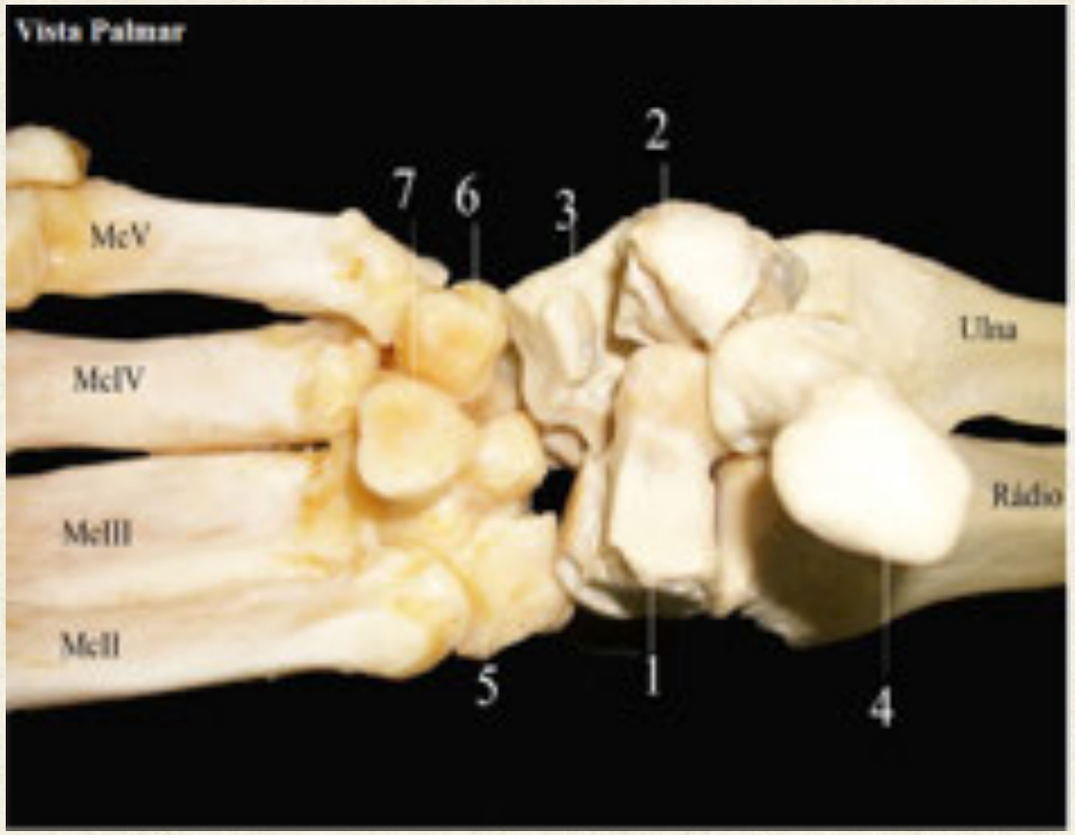


Figure 27 *– Carpal bones and metacarpal of T. terrestris , palmarl view. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: (1) Carpi radialis; (2) Carpi intermedium; (3) Carpi ulnare; (4) Carpi accessorium; (5) Carpale II; (6) Carpale IV; (7) Carpale III; (McII) Metacarpale II; (McIII) Metacarpale III; (McIV) Metacarpale IV; (McV) Metacarpale V.

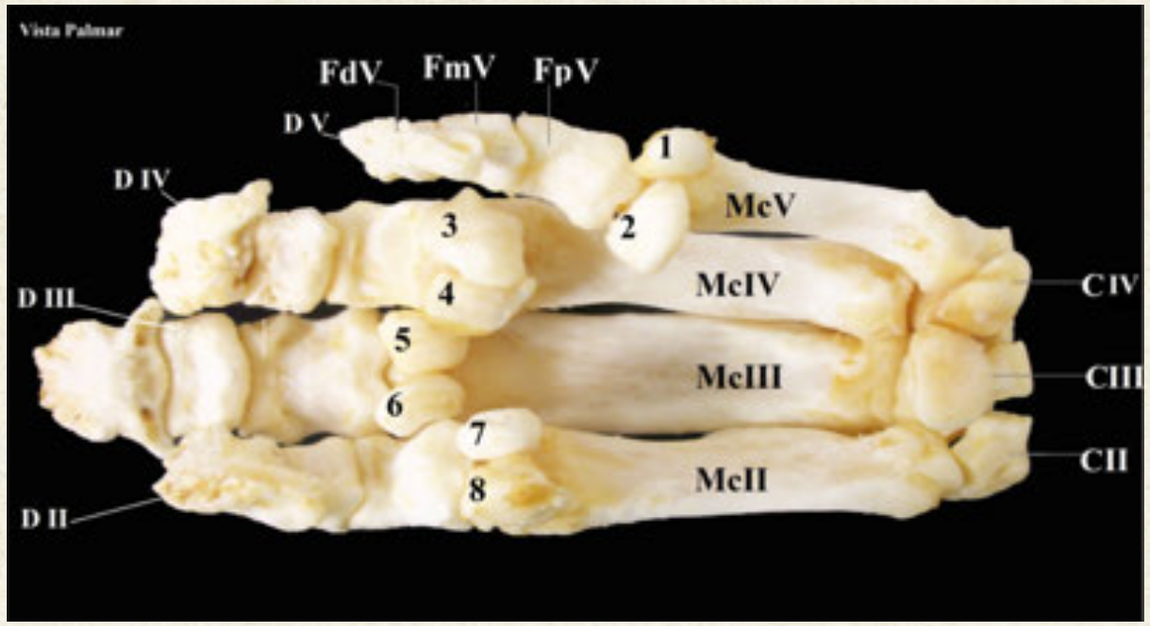


Figure 28 *– Hand bones of T. terrestris , palmar view. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: (CII) Carpale II; (CIII) Carpale III; (CIV) Carpale; (McII) Metacarpale II; (McIII) Metacarpale III; (McIV) Metacarpale IV; (McV) Metacarpale V; (1) e (2) Sesamóides dedo V; (3) e (4) Sesamóides dedo IV; (5) e (6) sesamoideum digiti III; (7) e (8) sesamoideum digiti II; (FpV) Phalanx proximalis digiti V; (FmV) Phalanx médialis digiti V; (FdV) Phalanx distelis digiti V; (D II) Digiti 2; (D III) Digiti 3; (DIV) Digiti 4; (DV) Digiti 5.

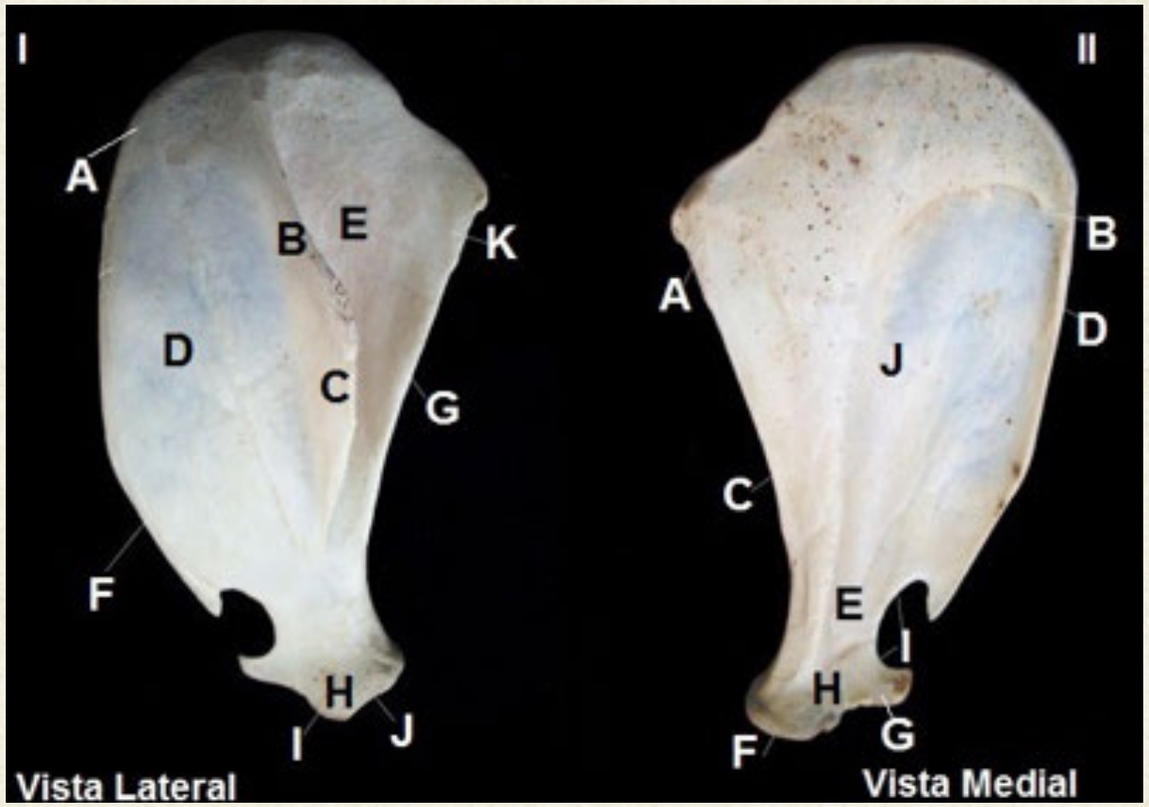


Figure 29 *– Scapula of T. terrestris (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: I: Lateral view: - (A) angulus cranialis; (B) spina da scapulae; (C) tuber scapulae; (D) fossa supraespinata; (E) fossa infraespinata; (F) margo cranialis; (G) margo caudalis; (H) foramem nutricia; (I) tuberculum supraglenoidale; (J) cavitas glenidalis; (K) angulus caudales.

II: Medial view - (A) angulus caudalis; (B) angulus cranialis; (C) margo caudalis; (D) margo cranialis; (E) colus scapulae; (F) cavitas glenoidialis; (G) tuberculum supraglenoidale; (H) processus coracoideus; (I) incisurae in margus lateralis distalis; (J) focies subescapularis.

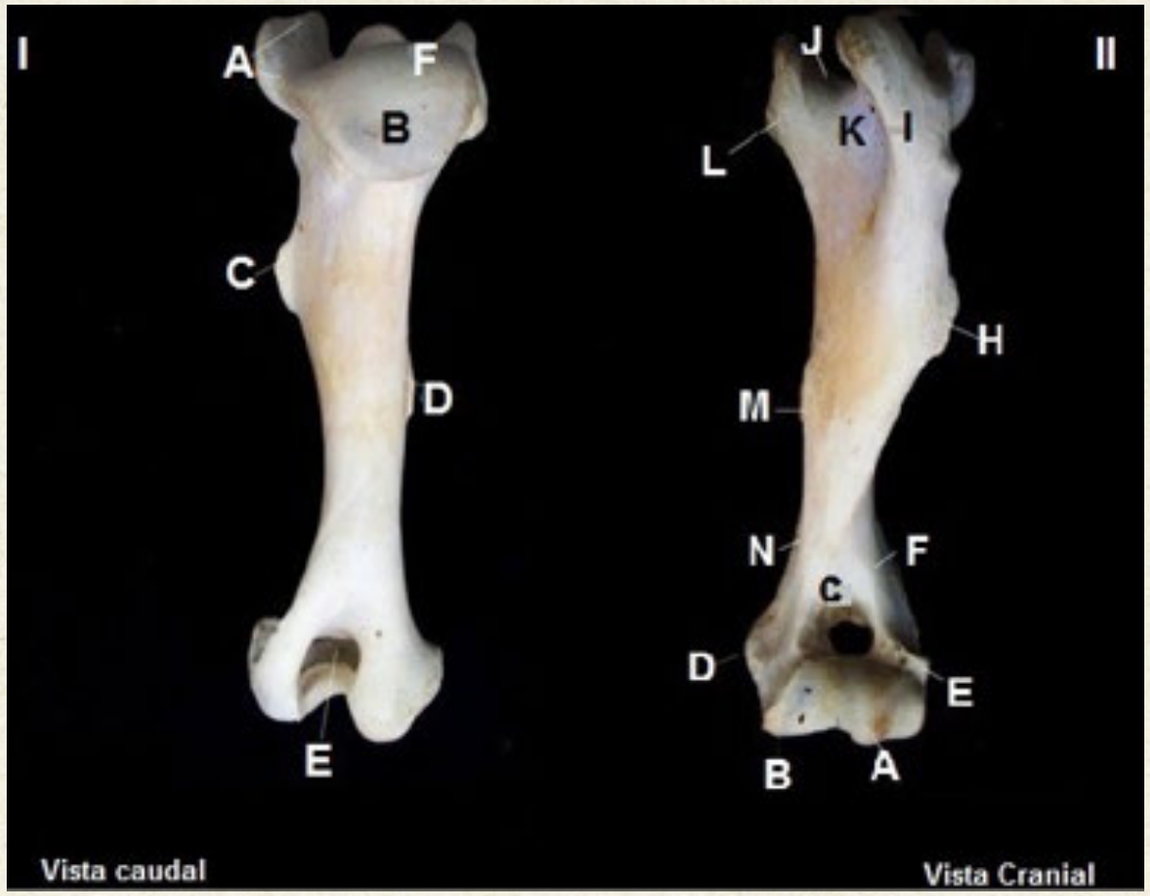


Figure 30 *– Humerus of T. terrestris. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legenda: I: Caudalis view - (A) tuberositas major; (B) colo; (C) tuberositas deltoidea; (D) tuberositas m. teres minor (E) focies olecrini (F) tuberositas humeri.

II: Cranialis view - (A) capitulum; (B) trochlea; (C) focies radialis; (D) epidicondylus medialis; (E) epicondylus lateralis; (F) crista epicondylus lateralis; (G) sulcus m braqui (H) tuberositas deltoidae; (I) tuberositas major; (J) sulcus intertuberalis; (K) foramem nutricia proximalis; (L) tuberositas minor; (M) tuberositas m. teres major; (N) foramem nutricia distalis.

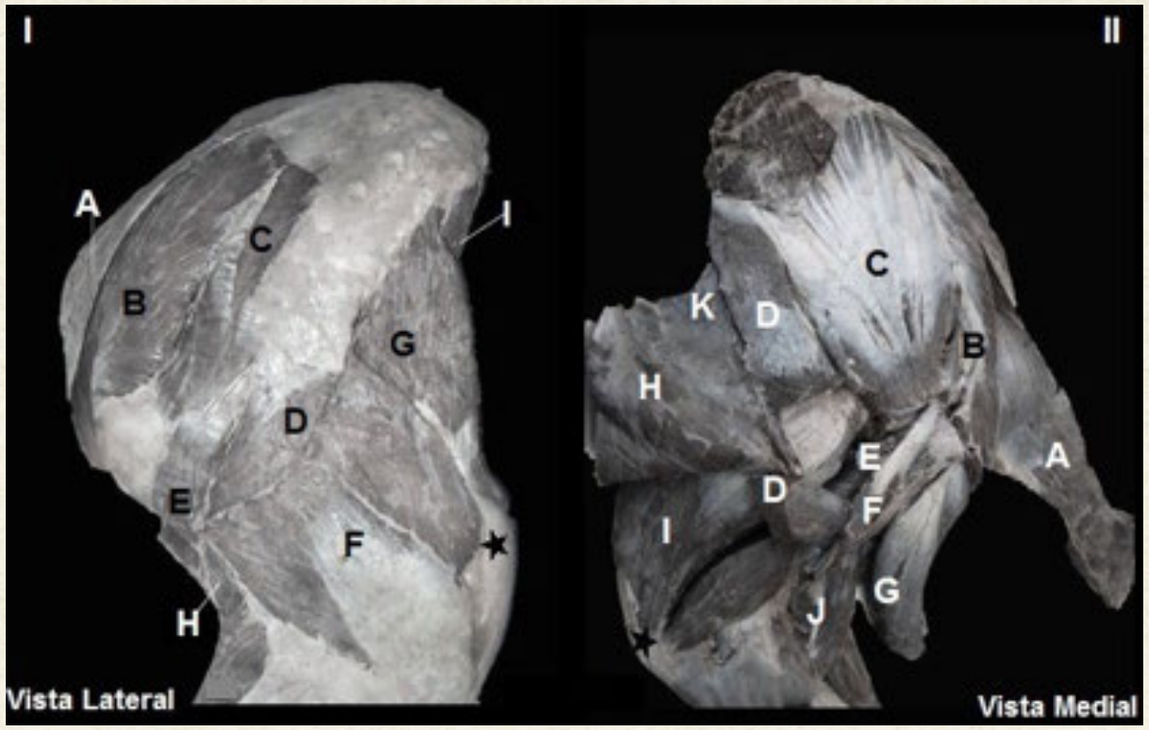


Figure 31 *– Musculus do cingulate of scapula of arms de T. terrestris, lateral and medial sides, respectively. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: I: Face side- (A) m. subclavius; (B) m. supraespinatus; (C) m. infraespinatus; (D) m. deltoideus; (E) m. teres minor; (F) m. triceps brachii caput laterale; (G) m. triceps brachii caput longum; (H) m. brachii; (I) m. tensor da fasciae antebrachii. II Face Medialis - (A) m. subclavius; (B) m. supraespinatus; (C) m. subescapularis; (D) m. teres major [partially folded]; (E) m. articularis Sholder; (F) m. coracobrachialis; (G) m. biceps brachii [partially folded]; (H) m. tensor da fascie do antibrachii [partially folded]; (I) m. triceps brachii caput mediale; (J) m. brachiallis; (K). m. large dorsalis [partially folded]; olecrani.

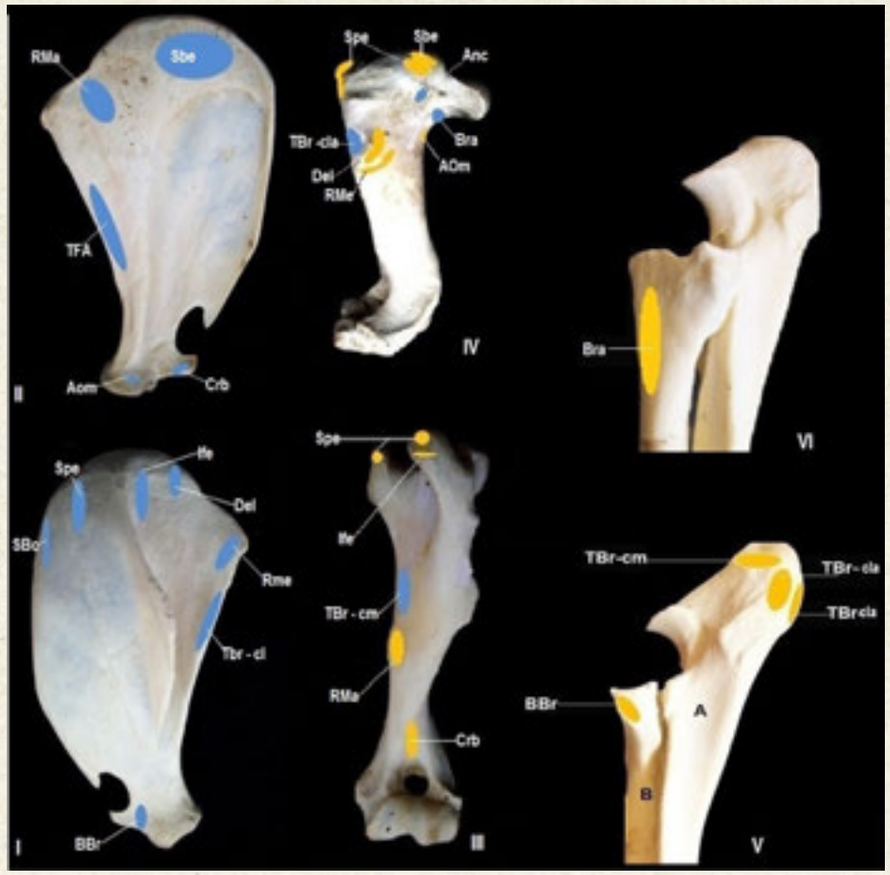


Figure 32 *– Fixing points of the scapular girdle and arm muscles Tapirus terrestris. Yellow: point of muscle insertion . Blue : point of muscular origin. (Fonte: PEREIRA, BORGES, SANTOS, 2013 - LAPAS).*

Legend: I: Side view of the scapula – (BBr) m. biceps brachii; (Del) m. deltuideus; (Ife) m. infraespinatus; (RMe) m. teres minor; (Sbc) m. subclavius; (Spe) m. supraespinatus; (TBr-cl) trceps brachii caput longus. II: Medial view of the scapula – (AOm) m. articularis shoulder; (Crb) m. coracobrachialis; (RMa) m. teres major; (Sbe) m. subescapularis; (TFA) m. tensor da fasciae antebrachii.. III: Cranial view of Humero – (Crb) m coracobrachialis; (Ife) m. infraespinhatus; (RMa) m. teres major; (Spe) m. supraespinhatus; (TBr-cm) m. triceps brachii caput mediale. IV: Side view of humero – (Anc) m. anconeus; (AOm)m. articularis shoulder; (Bra) m. brachiil; (Del) m. delteideus; (RMe) m. terer minor; (Sbe) m. subescapularium; (Spe) m. supraespinhalis; (TBr-cla) m. tríceps do braço cabeça lateral. V: Vista lateral da ulna (A) e rádio (B) – (BBr) m.biceps brachii; ((TBr-cl) m. triceps brachii caput longa; (TBr-cla) m. triceps brachii caput laterali; (TBr-cm) triceps brachii caput mediale. VI: View Mediale ulna – (Bra) m. brachii.

Newborn tapirs' coats have black or dark brown lines and white spots. They are completely white on the belly, chest and thorax. This color pattern is the same in all four species. Within three to four months the lines and white spots begin to fade; between eight and nine months the white patterns on newborns have disappeared from several parts of the body and those remaining are barely visible. At approximately year of age, juvenile tapirs have the same hair color pattern as adult specimens. Newborn tapirs usually weigh from 3-6 kg (Padilla & Dowler 1994). In captivity, lowland tapir calves gain an average of 2.5 kg per week and are completely weaned at 4 months of age (Barongi 1993). Growth is usually completed by 18 months (Young 1961).

Reproductive anatomy of female and male tapirs is described in Chapter 9 of this manual.

Figure 33 *- Lowland tapir. Newborn coat. Photo: Daniel Zupanc*

Figure 34 *- Lowland tapir. Adult coat. Photo: Daniel Zupanc*

RECOMMENDED LITERATURE

Borges DCS. 2013. Anatomia óssea e muscular e aspectos adaptativos do membro pelvino de Tapirus terrestris (Perissodactyla, Tapiridae). MSc Dissertation. Laboratório de Pesquisas em Animais Silvestres, Faculdade de Medicina Veterinária, Universidade Federal de Uberlândia (UFU), Minas Gerais, Brazil. 73f.

Mangini PR. 2007. Perissodactyla - Tapiridae (Anta). In: Tratado de Animais Selvagens: Medicina Veterinária, Cubas ZS, Silva JCR, Catão-Dias JL, editors. Editora Roca, São Paulo, Brazil, pp.598-614.

Medici EP. 2010. Assessing the Viability of Lowland Tapir Populations in a Fragmented Landscape. PhD Dissertation. Durrell Institute of Conservation and Ecology (DICE), University of Kent. Canterbury, United Kingdom.

Medici EP. 2011. Family Tapiridae (TAPIRS). In: DE Wilson & RA Mittermeier (Eds.). Handbook of the Mammals of the World - Volume 2: Hoofed Mammals. Lynx Edicions, Spain.

Medici EP; Mangini PR; Fernandes-Santos RC. 2014. Health Assessment of Wild Lowland Tapir (Tapirus terrestris) Populations in the Atlantic Forest and Pantanal Biomes, Brazil (1996-2012). In: Journal of Wildlife Diseases 50(4):817-828.

Padilla M; Dowler RC. 1994. Tapirus terrestris. Mammalian Species, 481:1-8.

Pereira SG. 2013. Anatomia óssea e muscular e aspectos adaptativos do membro torácico de Tapirus terrestris (Perissodactyla, Tapiridae). MSc Dissertation. Laboratório de Pesquisas em Animais Silvestres, Faculdade de Medicina Veterinária, Universidade Federal de Uberlândia (UFU), Minas Gerais, Brazil. 79f.


Picture: Patrícia Medici

3 Capture Methods

Capture Methods

The process of capturing tapirs must be carefully planned, and capture techniques must be chosen to minimize stress and injuries to study animals, non-target species, and researchers. Safety must be a priority before, during, and after capture, including throughout biological sample collection, marking, radio-transmitter placement, transport, translocation, etc. Aside from safety, many other factors determine the most appropriate capture methods, such as the species to be captured, local environmental conditions, and the need and availability of personnel and equipment.

Whatever the method selected, the first step is to identify areas frequently used by tapirs, then pre-bait and monitor these locations. Effective baits can improve trapping efforts considerably. Mineral salt and/or native forest fruits have both been used successfully. However, the bait only serves as extra incentive to tapirs to return a specific area regularly. It is critical that these bait stations are placed in areas regularly used by tapirs, including tapir paths, patches of fruiting trees, or other areas with abundant tapir sign. In the Calakmul region (Mexico), bait did not enhance researchers tapir trapping efforts. In this location, the researchers baited locations close to water bodies with bananas and mineral salt and waited for tapirs in the trees closeby. This technique yields better results during the dry season when tapirs seek permanent water bodies (Jonathan Perez, personal communication). Attempts to bait tapirs were likewise unsuccessful in Nicaragua. Researchers set up bat stations on tapir trails and monitored them for over a year with camera traps. They also monitored tapir trails that were not baited. Tapir visitation rates were not statistically higher at sites with bait than at unbaited sites (Christopher Jordan, personal communication).

Depending on the density of the tapir population to be studied, the first step may not be an easy one. Nonetheless, choosing sites with high tapir traffic is extremely important to the success of the research. Prior local knowledge of tapir behavior and familiarity with the study area usually helps the researcher

determine areas of greater tapir traffic. The experience of local hunters and ranchers can be extremely useful. Once tapirs have been visiting a particular bait station regularly the researcher should make a decision about the most appropriate capture method for that particular area.

In order to capture and chemically restrain wild tapirs it is absolutely vital that the personnel involved is well-trained and prepared to operate as a team. Stress and trauma to researchers and captured animals are intrinsic risks of handling wild tapirs. However a well-planned capture method and the selection of an effective and safe chemical restraint protocol can significantly reduce these risks.

The process of capturing tapirs requires the presence of at least one wildlife veterinarian to be in charge of immobilizing the animals, monitoring the captured animal's health while anesthetized, and collecting and processing biological samples. The involvement of a veterinarian who can select and administer a safe anesthetic protocol and carefully monitor the animals during anesthesia will further reduce intrinsic risks of the capture process. Veterinarians are the only professionals qualified to quickly identify anesthetic depression and take the appropriate measures. Thus, any researcher planning on capturing and immobilizing tapirs in the wild MUST budget for the participation of a well-experienced wildlife veterinarian. In many countries, this is a legal requirement, as only experienced veterinarians or other trained personnel are given the legal authorization to handle anesthetics (Benoit Thoisy, personal communication).

3.1. Anesthetic Dart Shooting

Figure 35 *- Anesthetic dart shooting from the ground. Photo: Patrícia Medici*

In some instances, it is possible to capture tapirs by shooting the animals with darts containing anesthetic solutions, either from a platform built near a bait station or from the ground. Compressed air or CO2 powered projectors should be used. Powder systems are not recommended as they make a comparatively loud noise when discharged that can startle and unduly stress tapirs. The bait station should be up to 10 meters from the platform, and the setup must be designed to ensure a clear shot with minimal obstacles. The main advantage of the darting method is that it does not require any major constructions, which means that the selected capture site is fairly undisturbed. Lower cost when compared to other captures methods is another important advantage of darting.

The main disadvantage of this method is that darted tapirs are not physically restrained. There are some inherent dangers in darting and working with wild tapirs that are not confined. The physiologic effect of stress may delay anesthetic induction or even prevent an animal from becoming sedated enough to stop moving. Even in cases where the anesthesia is effective, if the animal becomes frightened from the dart impact, it may run, and can cover a lot of ground before the anesthetic drugs take effect. If the capture site were nearby or within swamp or marshy areas, this could put tapirs at risk of drowning. It is recommended to avoid darting tapirs in wetlands or inn close proximity to bodies of water. In all darting scenarios, it is strongly recommended to use transmitter darts in order to facilitate the process of following and locating darted tapirs.

If darting near water bodies is necessary, the use of camera traps can be helpful to check the weight and the body condition of the animals that visit regularly, so the veterinarian can calculate an effective dose of the drugs that will be administered and prepare anesthetic darts before capture attempts (Jonathan Perez, personal communication).

Darting from tree platforms was the main capture method used by Charles R. Foerster and Sonia Hernandez-Divers to capture Baird's tapirs in Corcovado National Park, Costa Rica (Hernández-Divers & Foerster 2001).

Researchers from the Lowland Tapir Conservation Initiative in Brazil darted tapirs primarily from the ground to capture/re-capture five (5) lowland tapirs in the Atlantic Forest (Medici 2010) and 20 in the Pantanal (EP Medici, personal communication, 2014).

Further details about this capture method are provided in:

Hernández-Divers SM; Foerster CR. 2001. Capture and Immobilization of Free-Living Baird's Tapirs (Tapirus bairdii) for an Ecological Study in Corcovado National Park, Costa Rica. In: Zoological Restraint and Anesthesia, D. Heard (Ed.). International Veterinary Information Service, Ithaca, New York, USA.

Medici EP. 2010. Assessing the Viability of Lowland Tapir Populations in a Fragmented Landscape. PhD Dissertation. Durrell Institute of Conservation and Ecology (DICE), University of Kent. Canterbury, United Kingdom.

3.2. Pitfall Traps

Pitfalls for capturing tapirs consist of holes in the ground covered with corrugated roofing tiles and camouflaged with forest debris. The traps must be unnoticeable, and the same animal can be captured repeatedly. Pitfalls used to successfully capture lowland tapirs in the Atlantic Forest of Brazil (Medici 2010) were 2.3 m long, 1.5 m wide and 2.2 m deep. These dimensions may not be adequate for other tapir species in other locations. Tapirs may be able to escape from pitfalls less than 2 m deep. It is important to emphasize that the pitfalls should be dug in frequently visited tapir paths. Pitfall traps do not require baiting, so they are an alternative to other methods in field sites where baiting tapirs has proven ineffective. In the event of a capture, the tapir must be immobilized and manipulated inside the pitfall. This allows researchers full control during the recovery and release processes. Animals captured in pitfalls usually remain calm, and researchers can easily dart them with a CO2 powered pistol or a blowpipe and anesthetic darts. The impossibility of a tapir escaping while using this capture method allows for the design of safer protocols, with correct administration of pre-anesthetic drugs and capability to hold the animal until recovery is complete. Once the capture procedures are finished the veterinarian can then administer the reversal agents and monitor the recovery of the animal very closely. When the tapir is fully recovered from the anesthesia a ramp is opened into one of the sides of the hole and the animal is free to exit using the ramp. The biggest disadvantage of the pitfall trap is that once a tapir has been captured the same trap cannot be used again. The trap has to be closed. Other disadvantages of the pitfall traps are the difficulty and cost of digging such large holes. Groups of 4-5 people can take ~5-6 hours to dig and camouflage a pitfall. In addition, this technique can be controversial due to fracture hazard, the risk of catching more than one animal at a time, the risks of manipulation of the captured animal inside the hole, and the resulting habitat disturbance. Local geologic conditions and the local water table must be considered. In areas with a water table <2m, pitfalls should not be used or may only be a viable capture technique in the dry season.

This method was successfully used by the Lowland Tapir Conservation Initiative in Brazil to capture/recapture 15 lowland tapirs in the Atlantic Forest (Medici 2010). In addition, this method was recently used by researchers of the Nicaraguan Tapir Project to capture Baird's Tapirs in that country (C. Jordan, personal communication, 2014).

Figure 36 *- Pitfall traps building. Photos: Patricia Medici*

Further details about this capture method are provided in:

Medici EP. 2010. Assessing the Viability of Lowland Tapir Populations in a Fragmented Landscape. PhD Dissertation. Durrell

Figure 37 - *Anesthesia administration and manipulation of a lowland tapir inside a pitfall trap. Photos: Patrícia Medici.*

3.3. Capture Pen (Corral or Box Trap)

Capture pens consist of 3.5 m long, 1.5 m wide and 2.2 m high wooden enclosures. The posts of the traps (6) should be wider than 10 cm and the wooden boards thicker than 2.5 cm. The walls should be at least 2.2 m high to ensure tapirs cannot escape. These enclosures are equipped with a trigger installed in the back of the trap that holds the main trap door in place. When a tapir steps on or kicks the trigger, the trap door is automatically released, capturing the animal inside.

As with prior techniques, it is necessary to build capture pens on natural tapir paths or other areas that tapirs frequently use, such as salt licks, fruiting trees, or palm patches. A locally effective bait should be placed inside traps to attract the tapirs. In the event of a capture, the tapir is immobilized and manipulated inside the trap. Tapirs are immobilized through the use of CO2 powered pistol, or blowpipes, and anesthetic darts. Once the capture procedures are finished the veterinarian can then administer the reversal agents and monitor the recovery of the animal very closely. Once the tapir has fully recovered from the anesthesia, the door of the trap is opened and the animal is free to exit the trap through the door. Similar to the case with pitfalls, the main advantage of capture pens is that the team can manipulate tapirs inside the trap and has full control of the recovery and release processes. Another important advantage of this method is that despite the high initial effort and cost to build the traps, many different individual tapirs from the same area can be captured in the same trap. This reduces capture costs per tapir over the long-term and maximizes chances to collect data about intra-specific interactions between neighboring individuals. Capture pens used in the long-term must be regularly repaired.

This method was successfully used by the Lowland Tapir Conservation Initiative in Brazil to capture/re-capture 23 lowland tapirs in the Atlantic Forest (Medici 2010) and 114 lowland tapirs in the Pantanal (EP Medici, personal communication, 2014).

Gallery 4 - Box trap

Two different styles of capture pens. Note that the first trap was built in a patch of palm known to be an important food source for lowland tapirs. Photos: Renata Carolina Fernandes-Santos.

Further details about this capture method are provided in:

Medici EP. 2010. Assessing the Viability of Lowland Tapir Populations in a Fragmented Landscape. PhD Dissertation. Durrell Institute of Conservation and Ecology (DICE), University of Kent. Canterbury, UK.

Figure 38 *- Tapir being caught in a box trap (Camera trap photos).*

Picture: Bill Konstant

4

Chemical Restraint

Chemical Restraint

Several anesthetic protocols have been previously developed and used for captive (Janssen et al. 1999; Nunes et al. 2001; Janssen 2003) and wild tapirs (Hernandez et al. 2000; 2001; Mangini et al. 2001; Mangini 2007). The specific protocol used will vary depending on a variety of factors, including: capture method, goals of immobilization, desired or feasible drug volume, desired time for induction and recovery, required level of immobilization and muscle relaxation, need for reversibility, project budget, and requirements for safety of the animal and the members of the field team.

Several tapir field projects have used anesthetic protocols which have been exhaustively tested in wild tapirs in different areas (Parás-García et al. 1996, Hernández-Divers et al. 1998; Hernández-Divers et al. 2000; Hernández-Divers & Foerster 2001; Mangini et al. 2001; Mangini 2007; Medici 2010; Pérez 2013). Some of these protocols are presented below.

Researchers planning on using these protocols are strongly advised to consult with an experienced wildlife veterinarian prior to implementing the protocol in the field. In addition, it is highly recommended that the veterinarians who developed and/or have experience with each of these protocols be contacted for further consultation.

The conditions under which these protocols are successful should be carefully explored and considered when attempting to apply them to different situations.

Further information about the drugs described in this chapter and their effects on animal physiology are available in APPENDIX 1.

4.1. Important aspects to be considered

• The success of the chemical restraint of wild and captive tapirs depends on careful planning. During the planning stages, it is important to consider the following:

1.Characteristics of the anatomy, metabolism and physiology of the species;

2.The environmental conditions of the location where the capture will take place;

3.The capture method that will be employed;

4.The available equipment that might be used during the capture process;

5.Detailed knowledge of the pharmacology, adverse effects and counter-indications of the drugs that will be used for the chemical restraint;

6.Estimates of the time required to carry out all the procedures during the manipulation of the animal, including but not limited to: radio-tagging, installation of microchip, collection of biological samples, and clinical evaluation;

7.The possibility of unexpected events interrupting or interfering with the chemical restraint.

8.The need to check different physiological parameters of the tapirs during their recovery from anesthesia.

• It is not possible to easily and accurately determine the exact weight of a captured wild tapir. In some contexts, it can also prove difficult in captive animals. As calculating the weight is important to determine the proper dose of anesthesia, it is important to choose protocols that allow veterinarians to make estimates with a wide safety margin. Given the considerable weight of tapirs, the calculation of predetermined doses for body weight estimates at 50 Kg intervals is usually safe. If possible, we recommend that field personnel practice calculating tapir weights with captive animals of known weight. In the wild, camera traps can be used to help estimate individual tapir weights and/or to ensure that animals to be captured exhibit good body condition. Both of these observations can help veterinarians calculate the doses of drugs.

• The chemical restraint procedure should be performed during the cooler hours of the day. The animal must be closely and carefully monitored until it has fully recovered. After the manipulation, the animal should be capable to perform all of its ecological functions before its release.

• During the capture and immobilization procedures, it is important to minimize noise and recomendable to limit the field team to a small, manageable number of necessary personnel. As soon as the animal falls under the effect of the anesthesia, its eyes and ears must be covered in order to minimize external stimuli. This is particularly the case when dissociative anesthetic agents are used.

• It is necessary to prepare protocols for possible emergencies (see the most common emergencies in the following topics) and therapeutic protocols in advance (see details in Chapter 11). It is recommended to have emergency drugs available, examples include anesthetic antagonists such as Doxapram, Atropine, and Epinephrine.

Figure 39 *- Intramuscular administration of anesthetic agents in lowland tapir. Photo: Renata Carolina Fernandes-Santos*

• The intramuscular administration of anesthetic agents can be applied on the side of the neck or on the gluteal or thigh musculature (recommended for darting).

• Once the anesthetic agents start to take effect, whenever possible, the head of the tapir should be positioned below the level of the body to avoid aspiration in the case of regurgitation. It is advisable that veterinarians avoid the conditions that would lead to aspiration of gastric reflux. Tracheal tubing is difficult in tapirs due to their anatomy: the head is long and narrow and the glottis is not visible. Blind intubation is possible with experience. Tracheal tubes must be 10-14 mm for juveniles and 16-24 mm for adults. Direct observation of the larynx is possible with a long laryngoscope blade. In the case that the veterinarian doesn't have a tracheal tube it is very important that all the airways are clean. To be certain of this, he/she must take out the tongue and check the position of the animal to avoid obstructions.

Figure 40 *- Induction: lowland tapir showing firsts signs of sedation. Photo: Patrícia Medici*

• When dealing with wild animals, it is generally impossible to have a proper clinical evaluation prior to the restraint. Most of the times, it is only possible to have a rough evaluation of body condition, skin lesions and deformities by visual inspection. The conditions of the circulatory and respiratory system will be unknown until the animal is already chemically immobilized. This is a significant risk and veterinarians must

be aware of possible associated complications during anesthetic and chemical restraint procedures.

• The handling of extremely stressed animals should be avoided. Acute stress can have serious effects on animal response to natural threats, and their cardio respiratory system and metabolism, which can alter the effect of anesthetic agents and even put the animal's life at risk.

Figure 41 *- Monitoring physiological parameters of a wild lowland tapir under anesthesia. Photo: Gabriel Damasceno.*

• It is important to make sure that during the anesthetic induction or recovery, the animal can not access water or rocky or uneven terrain. Such conditions can lead to severe injury or even lethal accidents for anesthetized animal.

• The accessibility to the animal (depending on the selected capture method) and the volume of anesthetics to be administered are key parts of the planned protocol that will help determine the most appropriate equipment to administer the drugs (syringe, dart pistol, blowpipe, rifle etc.). For darting expeditions, dart pistols or rifles and special anesthetic darts can be purchased from well-known manufacturers such as Dan-Inject, Telinject, Pneu-Dart, etc. For animals in box-traps or pitfalls, blowpipes, injection sticks or syringes can be used.

• The ideal anesthetic protocol will be effective in a single dose, will force the animal to fall asleep promptly, and will provide sufficient time for all the necessary procedures. The protocol should be designed such that a veterinrian can easily and safely supplement it by administering additional drugs if there is a need to extend the handling period.

• Some anesthetic and/or reversal agents are either not available or illegal in certain countries. For instances protocols that include opioids are not legal in all tapir range countries. In countries in which anesthetic agents are banned, it may be necessary to develop alternative protocols for the chemical restraint of tapirs that those listed in the present manual. If this is necessary, these alternative protocols must be tested by qualified personnel under well-defined research designs before implementing them in the field.

Figure 42 *- Intravenous administration of reversal agents in a lowland tapir. Photo: Patrícia Medici.*

• The most common adverse effects during the induction or recovery from chemical restraint in tapirs are apnea, arterial hypotension and agitation/ataxia.

• The most common emergencies during tapirs captures are hypothermia, hyperthermia, bradycardia low oxygen saturation and apnea. The continuous monitoring of body temperature is essential to the safety of the immobilized animal given that drugs tend to interfere with thermoregulation functions. It is important to pay extremely close attention to body temperature on notably cold or hot days. The immobilized animal should not be exposed to cold air streams, wet surfaces, direct sun or environments with poor air circulation and increased temperature. Due to their large body mass and low body surface area to body mass ratio, tapirs are more prone to develop hyperthermia than hypothermia. Animals in hypothermia should be exposed to heat and/or protected with thermo isolants, while animals in hyperthermia should be bathed with fresh water.

• It is necessary to carefully monitor the physiological parameters of the animal under anesthesia. Careful auscultation of the heart and lungs; constant monitoring of heart and respiratory rates, body temperature, and mucosa coloration; and indirect blood pressure measurements (such as capillary refill time) are the basic parameters that must to be monitored. The respiratory rate, type and amplitude are the most important parameters to monitor anesthetic depression in tapirs. Monitoring of blood oxygen saturation with pulse oxymetry is also recommended, especially with anesthetic protocols that may involve episodes of short apnea. It is important to keep in mind that tapirs are adapted to experience physiological apnea while swimming. Thus short periods of apnea during the chemical restraint tend to be less compromising for these species.

• The veterinarian in charge of a tapir capture and chemical restraint should be fully acquainted with the physiology of stress and the potential medical consequences of capturing a wild animal. A species' stress level is one of the most important factors affecting the response to anesthesia in wild animals. Different tapir species and even different individuals of similar body weights can respond differently to the same type of stressful events, and thus to the same anesthetic protocol. All captures should be carefully planned to reduce stress for the captured animal. Again, this includes reducing noise and other stimuli to which the captured animal will be exposed as much as possible.

- It is preferable to use anesthetic drugs for which there is a reversal drug. The use of reversal drugs makes capture expeditions more efficient and reduces the time that researchers spend processing each animal. This can allow for safer captures in less than ideal conditions and make it feasible for a team to capture and process multiple animals in a single day or night.

Figure 43 *- Recovery/release. Photo: Patrícia Medici*

It is essential to keep a detailed record of the anesthetic doses and physiological monitoring during each capture. The results of these records, their success and failures must be published or somehow made available to other field researchers, to help improve our knowledge on the chemical restraint of tapirs. APPENDIX 2 presents a model of spreadsheet to record and monitor chemical restraints in the field.

It is essential to keep a detailed record of the anesthetic doses and physiological monitoring during each capture. The results of these records, including the positive and negative aspects must be published or somehow made available to other field researchers to help improve our knowledge on the chemical restraint of tapirs. APPENDIX 2 presents a model spreadsheet that can be used by field veterinarians to record and monitor pertinent details and field observations during chemical immobilization of wildlife.

In captivity it is quite common to capture or immobilize tapirs for a variety of activities, including veterinary examinations, radiographs, ultrasounds, the collection of biological samples, medical emergency, and transportation to a different enclosure. The type of management activity and the temperament of the individual tapir will determine if tapirs need to be immobilized or if operant conditioning will be adequate to perform required tasks.

We provide suggested protocols for those cases in which the anesthesia is required for captive tapirs (see Recommended Protocols in this Chapter). Anesthetizing captive tapirs is as delicate an operation as it is with wild tapirs. It is important that the veterinary staff is trained and experienced in capture or immobilizations to ensure the process goes as smoothly as possible. If capture equipment is used, it is important to ensure it is in good working condition.

Before anesthesia it is recommended hold the individuals without foot and water for 18-24 hours to reduce the risk of regurgitation and aspiration (this is rare, but not without precedent). The anesthesia procedures must be performed in a quiet environment (without shouting, noises or presence of other animals that might disturb the anetshetized tapirs). If the tapir that is to be immobilized shares an enclosure with

other tapirs, it is recommended to separate the animals before anesthesia to reduce risks for the tapirs and the veterinary staff.

Lateral recumbence is a comfortable body position for tapirs and convenient for the keeper and veterinary staff who work with the anesthetized animal. As with wild tapirs, the head and neck of the tapirs should be positioned below the level of the body to avoid aspiration of gastric reflux in case of regurgitation. Maintenance of venous access with fluid and supplemental oxygen via mask or nasal cannulation at 6–10L/min is recommended. During immobilization, it is important to cover the eyes with a cloth to avoid light stimulations. Similar of the case with wild tapirs, during the whole anesthesia process of captive tapirs, it is important for veterinarians to monitor important parameters such as heart rate, respiratory rate, body temperature and blood oxygen level.

In the case of captive tapirs, certain zoos perform operant conditioning (with positive reinforcement), making chemical immobilization unnecessary. In these cases, tapirs collaborate voluntarily throughout the process of different medical procedures or other management activities. When tapirs are scratched and rubbed along the dorsum, abdomen, neck, rostrum, or jaw line, they lie down. With particularly docile individuals, veterinarians can perform medical examinations, venipuncture, ultrasound, treatment of wounds, etc. while assistants scratch the tapir down. The people who work with trained tapirs must practice caution in order to prevent injuries or serious accidents (Janssen, 2003).

Gallery 5 - Chemical Restraint

Anesthesia in captive tapir. Reserva Experimental Horco Molle, Tucumán Province. Argentina. Photo: Temaiken Foundation.

Anesthesia recovery. Reserva Experimental Horco Molle, Tucumán Province. Argentina. Photo: Temaiken Foundation.

RECOMMENDED LITERATURE

Gachen G; Quse V; Falzone M; González Ciccia P. 2011.Effective drug combinations for collecting samples in lowland tapirs (Tapirus terrestris).In: Proceedings of the Fifth International Tapir Symposium. p32.

Hernández-Divers SM; Bailey JE; Aguilar R; Loria DL; Foerster CR. 1998. Cardiopulmonary Effects and Utility of a Butorphanol-Xylazine-Ketamine Anesthetic Protocol for Immobilization of Free-Ranging Baird's Tapirs (Tapirus bairdii) in Costa Rica. In: Proceedings of the American Association of Zoo Veterinarians (AAZV).

Hernández-Divers SM; Bailey JE; Aguilar R; Loria DL; Foerster CR. 2000. Butorphanol-Xylazine-Ketamine Immobilization of Free-Ranging Baird's Tapirs in Costa Rica. In: Journal of Wildlife Diseases: 36(2), pp. 335–341

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Parás-García A; Foerster CR; Hernández-Divers SM; Loria DL. 1996. Immobilization of Free Ranging Baird's Tapir (Tapirus bairdii). In: Proceedings American Association of Zoo Veterinarians (AAZV).

Pérez J. 2013. Immobilization of Baird´s Tapir (Tapirus bairdii) using thiafenthanil oxalate (A3080) in combination with xylazine and ketamine. In: Tapir Conservation - The Newsletter of the IUCN/SSC Tapir Specialist Group, vol. 22 (31), pp, 15-19.

4.2. Recommended Protocols

Basically, the anesthetic protocol includes a mix of opioid agonist (Butorphanol at 0.15-0.4 mg/kg IM) and an alpha-2 adrenergic agonist (Xylazine at 0.3-0.8 mg/kg IM or Detomidine at 0.05 mg/kg IM). These drugs can be administered in the same dart (eg. Butorphanol + Xylazine). Approximately 4-6 minutes after being darted, tapirs start with lack of coordination; they stagger and begin to sit down. After approximately 10 minutes there is a higher level of muscle relaxation and the animal lies down. If the initial anesthesia is not sufficient, Ketamine (0.25–1mg/kg IV) can be used for further restraint as needed (Janssen, 2003). The intramuscular administration of anesthetic agents can be applied on the side of the neck or on the gluteal or thigh musculature.

Alpha-2 and narcotic antagonists should be used as reversal agents (e.g. Yohimbine at 0.3 mg/kg IV and Naltrexone, at 0.6 mg/kg respectively) (Janssen, 2003). The recovery generally starts 1-2 minutes after administering the antagonist to the immobilized animal. In those cases in which the researcher supplemented the initial anesthesia with Ketamine, it is necessary to wait 30-40 minutes after the last administration of Ketamine before administering the reversal agents.

As mentioned above, during the anesthetic induction and recovery period, the captured tapir should not have access to pool water, rocky areas or habitats with steep inclines to avoid accidents (Nunes et al. 2001).

Butorphanol / Xylazine

Baird's *TapirTapirus bairdii* - Corcovado National Park, Costa Rica

Capture Method: Darting

Protocol: A total dosage for a 200-300 kg animal was comprised of 40-50 mg of Butorphanol Tartarate (Torbugesic) plus 100 mg of Xylazine in the same dart. Additional Ketamine 187±40.86 mg/animal, administered IV the majority of times, to maintain/extend the anesthesia.

Reversal: Naltrexone (50 mg) mixed with 1200 mg of Tolazoline in the same syringe, IM, given no sooner than 30 minutes from the last administration of Ketamine.

Comments: This protocol was administered to animals from a tree blind via dart. The animals had been habituated to come to bait (ripe bananas) for several days and thus were relatively calm when darted.

For further information about this protocol please contact:

Charles R. Foerster (Researcher), Sonia Hernández (DVM; E-mail: shernz@uga.edu)

Further details about this protocol are provided in:

Hernández-Divers SM; Bailey JE; Aguilar R; Loria DL; Foerster CR. 1998. Cardiopulmonary Effects and Utility of a Butorphanol-Xylazine-Ketamine Anesthetic Protocol for Immobilization of Free-Ranging Baird's Tapirs (Tapirus bairdii) in Costa Rica. Proceedings of the American Association of Zoo Veterinarians (AAZV).

Hernández-Divers SM; Bailey JE; Aguilar R; Loria DL; Foerster CR. 2000. Butorphanol-Xylazine-Ketamine Immobilization of Free-Ranging Baird's Tapirs in Costa Rica. In: Journal of Wildlife Diseases: 36(2), pp. 335–341

Hernández-Divers SM; Foerster CR. 2001. Capture and Immobilization of Free-Living Baird's Tapirs (Tapirus bairdii) for an Ecological Study in Corcovado National Park, Costa Rica. In: Zoological Restraint and Anesthesia, D. Heard (Ed.). International Veterinary Information Service, Ithaca, New York, USA.

Butorphanol / Medetomidine / Ketamine
Lowland Tapir *Tapirus terrestris* - Pantanal, Brazil
Capture Method: Capture Pen

Protocol: Butorphanol Tartarate (Torbugesic, 10mg/ml) 0.15 mg/kg + Medetomidine (Domitor, 1mg/ml) 0.012 mg/kg + Ketamine (100mg/ml) 0.6 mg/kg, IM, in a 5cc dart. The concentrated version of Medetomidine (20mg/ml) can been used to reduce final volume. Atropine (15mg/ml) 0.03mg/kg can be added to the protocol in order to inhibit excessive salivation and respiratory secretions, commonly observed in lowland tapirs.

Reversal: Naltrexone (50mg/ml) 0.3 mg/kg + Atipamezole (Antisedan 5mg/ml) 0.04 mg/kg in the same syringe, ½ IM + ½ IV, given no sooner than at least 35 minutes from the last administration of Ketamine.

Comments: Adequate for tapirs captured in pens or pitfalls. The average induction time of this protocol is 4-6 minutes, and the average recovery time is 1-2 minutes after reversal administration. This protocol is also commonly used on lowland tapirs in captivity.

For further information about this protocol please contact:

Patrícia Medici (Researcher; E-mail: epmedici@uol.com.br), Renata Carolina Fernandes-Santos (DVM; E-mail:renatacfsantos@gmail.com.br), Paulo Rogerio Mangini (DVM; E-mail: paulomangini@triade.org.br), Caio Motta (DVM; E-mail: mvcaiomotta@gmail.com).

Metadona / Detomidine / Ketamine
Lowland Tapir *Tapirus terrestris* – Atlantic Forest, Brazil
Capture Method: Capture Pen

Protocol: Metadona Cloridrate (Mytedom, 10mg/ml) 0.15 mg/kg + Detomidine (Dormiun V, 10mg/ml) 0.05 mg/kg + Ketamine (100mg/ml) 1-2 mg/kg, IM. Atropine (10mg/ml) 0.02mg/kg can be added to the protocol in order to inhibit excessive salivation and respiratory secretions, both commonly observed in lowland tapirs.

Reversal: Naloxone (Narcan 0.4mg/ml) 0.01 - 0.02 mg/kg + Yohimbine (10mg/ml) 0.1 mg/kg in the same syringe, ½ IM + ½ IV, given no sooner than at least 35 minutes from the last administration of Ketamine.

Comments: Adequate for tapirs captured in pens or pitfalls. The average induction time of this protocol is 5-6 minutes, and the average recovery time is 1-3 minutes of reversal administration. Good muscular relaxation, allowing move the animal inside box trap or transporting box.

For further information about this protocol please contact:

Andressa Gatti (Researcher), Maria Fernanda Naegeli Gondim (DVM; E-mail: mfgondim@yahoo.com.br), Eduardo Raposo Monteiro (DVM), Paulo Rogerio Mangini (DVM¸ E-mail: paulomangini@triade.org.br), Renata Carolina Fernandes-Santos (DVM; E-mail: renatacfsantos@gmail.com.br).

Tiletamine-Zolazepam / Medetomidine / Ketamine

Lowland Tapir *Tapirus terrestris*- Atlantic Forest and Pantanal, Brazil

Capture Method: Darting

Protocol: Tiletamine-Zolazepam (Zoletil 50, 250mg) 1.25 mg/kg + Medetomidine (20mg/ml) 0.006 mg/kg + Ketamine (100mg/ml) 0.6 mg/kg, IM, in a 3cc dart. To reduce final volume, Ketamine and Medetomidine are used to dilute lyophilized Tiletamine-Zolazepam. Atropine (15mg/ml) 0.03mg/kg can be added to the protocol in order to inhibit excessive salivation and respiratory secretions, both commonly observed in lowland tapirs. Considering fairly common adverse effects observed during the recovery using this protocol (see Comments bellow), some veterinarians indicate the administration of Midazolam (5mg/ml) 0.03mg/kg 30-40 minutes after the administration of Zolazepam to supplement the benzodiazepine effects and reduce undesirable effects of dissociative anesthetics. On the other hand, the Midazolam supplementation could extend the recovery time.

Reversal: Not available

Comments: Adequate for the darting capture method (smaller total volume). The average induction time for this protocol is 2-3 minutes, with good immobilization time of 30 to 40 minutes. Considering that it is usually impossible to estimate tapir weight prior to use the darting method, this protocol provides a wide margin of safety. This protocol was designed for lowland tapirs with estimated body weight of 200kg but has been used and provided safe and satisfactory chemical immobilization for individuals with body weight between 100-300 kg. Variations of this protocol using other Alpha-2 agonists such as Detomidine and Romifidine, with and without Ketamine, were tested and best results for immobilization, cardio-respiratory parameters and recovery were obtained with Medetomidine and Ketamine. Short apnea episodes were observed more frequently using Detomidine protocols. It is not uncommon to observe adverse effects, as agitation, head shaking, pedaling, short apnea episodes and and ataxia during the recovery using Tiletamine/Zolazepam protocols.

For further information about this protocol please contact:

Patrícia Medici (Researcher; E-mail: epmedici@uol.com.br), Renata Carolina Fernandes-Santos (DVM; E-mail:renatacfsantos@gmail.com.br), Paulo Rogerio Mangini (DVM; E-mail: paulomangini@triade.org.br), Caio Motta (DVM; E-mail: mvcaiomotta@gmail.com).

Further details about this protocol are provided in:

Mangini PR; Velastin GO; Medici EP. 2001.Protocols of chemical restraint used in 16 wild Tapirus terrestris. Archives of Veterinary Science 6:6-7.

Mangini PR. 2007. Perissodactyla - Tapiridae (Anta). In: Tratado de Animais Selvagens: Medicina Veterinária, Cubas ZS, Silva JCR, Catão-Dias JL, editors. Editora Roca, São Paulo, Brazil, pp.598-614.

Nunes LAV; Mangini PR; Ferreira JRV. 2001. Order Perissodactyla, Family Tapiridae (Tapirs): Capture Methodology and Medicine. In: Biology, Medicine and Surgery of South American Wild Animals, Fowler ME, Cubas ZS, editors. Ames, Iowa University Press, USA, pp.367-376.

Etorphine / Acepromazine
Baird's Tapir *Tapirus bairdii* - Mexico
Capture Method: Darting

Protocol: The total dosage for a 200-250 kg animal is a mixture of 1.96 mg Etorphine Hydrochloride plus 5.90 mg of Acepromazine Maleate in the same dart.

Reversal: Diprenorphine Hydrochloride (Revivon Large Animal, C/Vet limited) - 5.88 mg

Comments: This protocol has been designed for the particular conditions of the Sierra Madre of Chiapas, Mexico. This region has a highly accented topography with pronounced slopes of more than 60 degrees of inclination. For this reason, the induction time must be minimized, in order to avoid fatalities.

For further information about this protocol please contact:

Alberto Parás-Garcia (DVM), Iván Lira-Torres (DVM; E-mail: ilira_12@hotmail.com)

Further details about this protocol are provided in:

Parás-García A; Foerster CR; Hernández-Divers SM; Loria DL. 1996. Immobilization of Free Ranging Baird's Tapir (Tapirus bairdii). In: Proceedings American Association of Zoo Veterinarians (AAZV).

Thiafenthanil Oxalate (A3080) / Xylazine / Ketamine
Baird's Tapir *Tapirus bairdii* - Mexico
Capture Method: Darting

Protocol: The drug combination was done with Thiafenthanil Oxalate 1mg/100kg + Xylazine 1mg/kg + 0.5mg/kg IM provided in the same dart.

Reversal: Naltrexone 10mg/1mg of Thiafenthanil and Yohimbine 0.125 mg/kg IM

Comments: This protocol has been used in the Calakmul region of Campeche, Mexico. The combination of these three drugs provide a rapid induction and recovery times. Animals with a poor body condition can be immobilized for short or long periods of time, these drugs demonstrated to be safe for free ranging and captive animals.

For further information about this protocol please contact:

Jonathan Pérez Flores (DVM; E-mail: johnspf77@yahoo.com.mx)

Further details about this protocol are provided in:

Pérez J. 2013. Immobilization of Baird´s Tapir (Tapirus bairdii) using thiafenthanil oxalate (A3080) in combination with xylazine and ketamine. In: Tapir Conservation - The Newsletter of the IUCN/SSC Tapir Specialist Group, vol. 22 (31), pp, 15-19.

Butorphanol / Xylazine
Lowland Tapirs *Tapirus terrestris* in captivity - Argentina
Capture Method: Darting

Protocol: The drug combination was done with Xylazine 0.8mg/kg IM + Butorphanol 0.4 mg/kg IM provided in the same dart. In the case that it was necessary to deepen the anesthesia for some reason (such as surgical procedure or if the immobilized animal was being transferred) Ketamine IV (1mg/kg) or inhalated Isofluorane were administered.

Reversal: Yohimbine 0.36 mg/kg IV (for the Xylazine) and Nalthrexone 0.4 mg/kg IV (for the Butorphanol), given after 30-40 minutes from the last administration of Ketamine.

Comments: This protocol was performed in 40 animals (24 males and 16 females; 38 adults and two juveniles) from five Argentinean zoos. Induction time was 12 minutes. In general, the procedures took between 10-45 minutes with an average of 28 minutes.

For further information about this protocol please contact:

Gustavo Gachen (DVM; E-mail: ggachen@temaiken.org.ar); VivianaQuse (DVM; E-mail: vivianaquse@gmail.com);Martín Falzone (DVM; E-mail: mfalzone@temaiken.org.ar).

Further details about this protocol are provided in:

Gachen G; Quse V; Falzone M; González Ciccia P. 2011.Effective drug combinations for collecting samples in lowland tapirs (Tapirus terrestris). In: Proceedings of the Fifth International Tapir Symposium. p32.

Main anesthetic protocols used on European captive lowland tapirs

Synthesis of the main anesthetic protocols used on European captive lowland tapirs (Lowland tapir EEP Vet questionnaire 2014)

Butorphanol 0.15 mg/kg	+	Xylazine 0.3 mg/kg	+ If needed	Ketamine 0.5 - 1 mg/kg IM or IV
		Medetomidine 0.01 - 0.03 mg/kg		
		Detomidine 0.04 - 0.05 mg/kg		

Recommended protocol for loading and transport of tapirs in captivity

The following protocol is a tranquillization for loading and transport of tapirs in captivity. It is very practical as it can be administrated orally the day before the transport: Acepromazine 0.75 mg/kg + Diazepam 0.05 mg/kg PO. This protocol was used in Lowland and Malayan tapirs. After the administration, the animal will be quiet in next 48 hours. Oral gel of Detomidine has also been used successfully for a slight and short sedation (Ordonneau 2014).

Picture: Daniel Zupanc

5

Clinical Evaluation

Clinical Evaluation

The clinical evaluation of a captured specimen begins upon the first sighting of the animal inside the trap (or during the pursuit for capture) when the veterinarian assesses the apparent health of the tapir, including its nutritional condition, the condition of the skin and hair, locomotion ability and estimated body weight. In the case of apparently unhealthy animals, with several skin lesions, bad nutritional condition, evident difficulty in locomotion etc., the veterinarian should choose against chemically restrain the animal, unless an alternative anesthetic protocol with more appropriate drugs can be used to safely immobilize the animal.

Figure 44 *- Physical evaluation of wild lowland tapir inside a box trap by visual inspection. Photo: Patrícia Medici.*

Once anesthetized, each tapir must receive a complete physical examination including assessments or examinations of:

1) overall body condition;

2) condition of fur;

3) skin integrity (presence of lesions, scars and/or wounds, alterations in pigmentation);

4) anatomical cavities (ophthalmic, nasal, ear, oral – including dental evaluation, rectal);

5) palpation and auscultation;

6) musculoskeletal integrity and mobility;

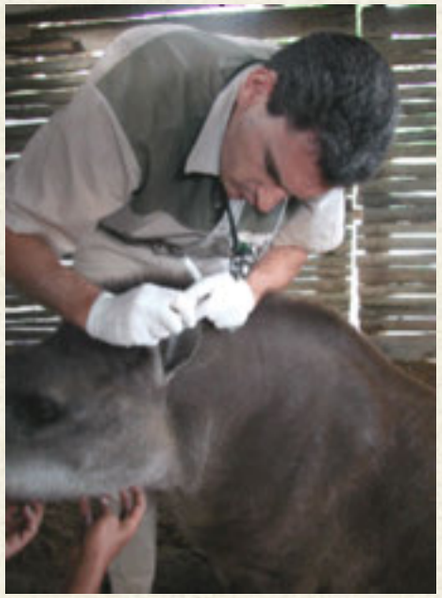
7) condition of nails and foot pads;

8) reproductive health (in females: vaginal inspection, evaluation of mammary glands and other evidence of reproductive activity; in males: evaluation of penis and palpation of testes); and,

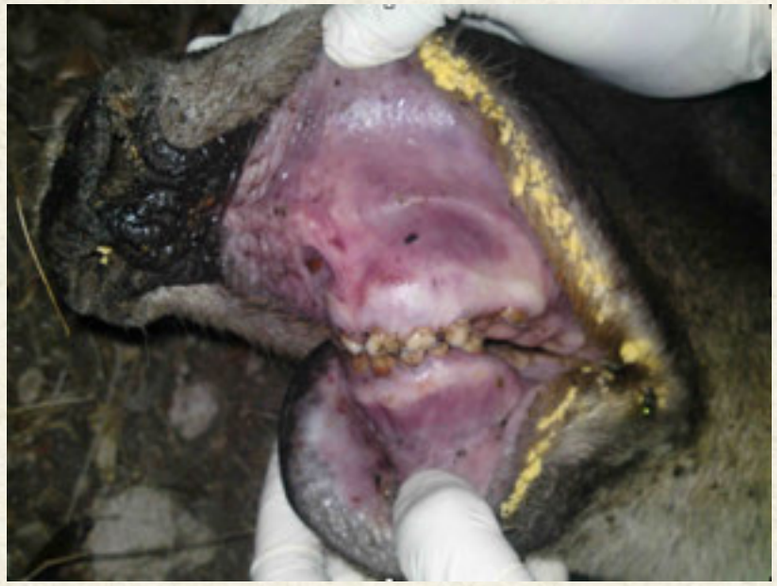
9) presence and level of infestation of ectoparasites (Medici et al. 2014).

Gallery 6 - Clinical Evaluation

Medical examination of wild tapir under anesthesia. Photo: Patrícia



Veterinary examination of captive tapir under operant conditioning. Photo: Temaiken Foundation.



Veterinary evaluation of oral mucosa in wild tapir. Photo: Jonathan Perez.

Table 3 *- Results of physical evaluation of wild lowland tapirs in the Atlantic Forest-AF (1996–2008) and Pantanal-PA (2008–2012), Brazil (Medici et al. 2014).*

Parameter	AF (N=44)	PA (N=68)
Body condition	77.3% good 15.9% regular 6.8% poor	75% good 23.5% regular 1.5% poor
Fur condition	6.8% altered pigmentation or physical hair condition	1.5% alopecia in the dorsal lumbar region
Skin	34.1% scars or recent wounds	47.1% scars or recent wounds 1.5% phlegmon
Eyes	9.0% senile halo 4.5% bilateral lens opacity 4.5% periophthalmic glandular inflammation	4.4% unilateral corneal lesions 2.9% senile halo 1.5% bilateral lens opacity 1.5% bilateral yellowish eye discharge
Genital	2.3% abnormal vaginal discharge 2.3% vaginal mucosal hyperemia	1.5% abnormal vaginal discharge
Dental conditions	4.5% fractures in incisors	No dental conditions observed

In many sites wild tapirs are heavily infested with ticks (mostly Amblyomma sp.), and also chiggers (Tunga penetrans). The data indicate that such infestations should be considered normal for many wild tapir populations. Nonetheless, whenever possible, the researcher should try to quantify the infestation, compare the degree of infestation with hematological parameters, and compare local results with those from different study sites in order to determine the impact of ectoparasites on local tapirs. Ticks and other ectoparasites tend to concentrate on the abdomen, ears, mammary glands, vulva/penis, inguinal region and medial side of the thigh.

During the clinical evaluation, captured individuals can be categorized according to age class based on dentition, teeth wear, and erosion of nails and appearance of foot pads. Medici (2010) separated individuals into three age classes: juvenile (6 months to 1 year), sub-adult (1-4 years), and adult (over 4 years of age). Younger individuals (less than 6 months) can be categorized as infants. Recording this information helps profile the population of captured tapirs and interpret health and spatial ecology data.

It has been reported that wild lowland tapirs wearing radio-collars for prolonged periods of time suffer local depletion on the crest and sometimes cutaneous deformations, alopecia and skin hardening under the collar. In some cases, the radio-collars cause chronic skin lesions by friction, which might predispose these individuals to local myiasis. If skin lesions are severe, tapirs must be recaptured and collars should be removed.

Figure 45 *- Lowland tapir showing local depletion on the crest and alopecia resulting from prolonged use of radio-collar. Photo: Patrícia Medici.*

It is necessary for veterinarians to perform periodic clinical evaluations on captive tapirs as well. Conditioned individuals that collaborate with medical management and procedures can be easily checked without immobilization. It might be necessary to anesthetize tapirs without training in order to evaluate their health. Anesthetization can afford veterinarians the option of collecting biological samples (blood, urine, etc.), or performing complementary exams such as ultrasound and radiography.

Table 4 *- Physiological parameters of wild lowland tapirs under anesthesia in the Atlantic Forest (AF) and Pantanal (PA) biomes, Brazil (Medici et al. 2014).*

Parameter		AF + PA		
		Mean	SD	N
Cardiac Rate	(bpm)	75	18	60
Respiratory Rate	(bpm)	26	10	59
Blood Oxygen Saturation	(%)	88	13	50
Body Temperature	(◦C)	37	1	32

SD = standard deviation

RECOMMENDED LITERATURE

Medici EP; Mangini PR; Fernandes-Santos RC. 2014. Health Assessment of Wild Lowland Tapir (Tapirus terrestris) Populations in the Atlantic Forest and Pantanal Biomes, Brazil (1996-2012). In: Journal of Wildlife Diseases 50(4):817-828.

Mangini PR; Medici EP; Fernandes-Santos RC. 2012.Tapir Health and Conservation Medicine. In: Journal of Integrative Zoology 7:331-345p.

Picture: Antwerp Zoo

6

Collection, Handling and Storage of Biological Samples

Collection, Handling and Storage of Biological Samples

There remains little available information on diseases affecting tapirs in the wild. The IUCN/SSC Tapir Specialist Group (TSG) has managed to encourage veterinarians and researchers working on tapirs to collect biological samples and share/publish results of their tapir health assessments. Thus, over the course of the last few years, we have been able to accumulate a great deal of information and are continuing to expand our knowledge of how diseases affect wild tapirs.

It is of utmost importance that veterinarians planning to collect biological samples consult with the diagnostic laboratory that will perform the analysis prior to the collection of samples to avoid inappropriate sample collection, handling or storage (Table X). Given that most commercially available diagnostic tests have been designed and tested for domestic animals, we strongly recommended that field veterinarians consult specialists in the different areas (microbiologists, virologists, etc.) to determine the appropriate test to use and understand how to accurately interpret results. In addition to this, we recommend that field veterinarians develop a system for longer-term storage in order to maintain samples for future analysis, as resources for new diagnostic tests are in constant development and more accurate or appropriate analyses may be available in coming years.

All biological samples collected should be traceable to the individual animal they were taken from (including information on species, name, sex, and age class). Furthermore, it is important to record: a description and the geographic coordinates of the location where the sample was collected, the season of collection (which might affect the prevalence of some diseases), a detailed history describing the conditions under which the samples were collected (sedation, general anesthesia, necropsy etc.), and any anatomical features (e.g. blood collection site, ectoparasite collection site) of the tapir that may help veterinarians interpret the results of diagnostic test results. APPENDIX 2 provides a suggested checklist of samples to be collected and notes to be taken. We strongly recommend the use of the spreadsheets provided to help standardize data collection globally and facilitate cross-site comparisons.

For tapirs in captivity, it is important to label the samples with the name of the institution/zoo, chip number, ZIMS identity or tapir's name, sex, age, date and time of collection and name of the person who collected the samples.

Finally, tapirs are listed by the Convention on International Trade in Endangered Species of Wild Fauna and Flora (CITES), meaning that the transport of any biological product deriving from them falls under CITES regulations. Whenever transporting samples out of the country of origin, in addition to other import/export permits, a CITES permit is required. The veterinarian is strongly encouraged to become familiar with the legislation in his/her country that regulates the movement of tapir biological samples both within and across national borders.

The following sections provide detail about the common biological sampling procedures.

6.1.1. Blood

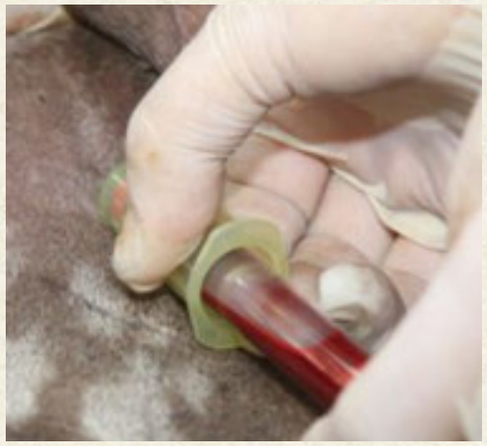
For each blood sample to be drawn, the area must be properly disinfected with 1:1 povidine iodine/ethanol 70% solution or chlorhexidine, given that tapirs are closely associated with water and their skin can be highly contaminated.

Venipuncture can be easily made in the saphenous or cephalic veins or in their carpal/tarsal derivates on medial access where the skin is thinner. The jugular vein is deep and not always easy to access, but is an important alternative when large blood volumes are necessary or when the other veins are collapsing after puncture. The caudal auricular vein, which runs along the center of the back of the ear, may also be used.

The use of vacuum sampling systems (e.g. Vacutainer®) is recommended for the collection of blood samples, as it avoids the contamination of the samples and allows collecting multiple samples from a single vascular puncture, reducing vascular trauma. With trained tapirs in captivity, the use of butterfly needles connected to a syringe or a vacuum sampling system can be very helpful, as this system enables slight movements of the animal without any vascular trauma.

It is recommended that blood samples be collected within 30 minutes of immobilization in order to minimize the influence of anesthesia on hematological parameters. Samples should be placed in a portable cooler and transported from the capture site to a field laboratory as soon as possible, then pre-processed and properly stored for later analyses in reference laboratories.

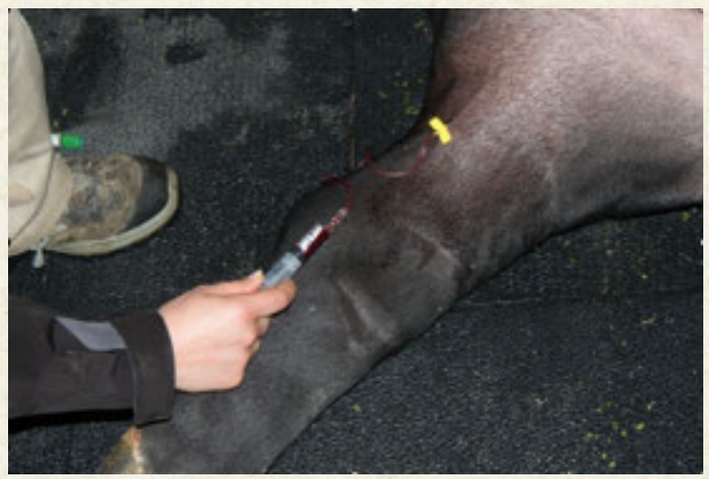
Gallery 7 - Blood collection



A) Vacuum sampling system. Photo: Patrícia Medici

B) Blood collection in captive tapir under operant conditioning. Photo: Temaiken Foundation

C) Blood sample collection in wild tapir under anesthesia. Photo: Patrícia Medici



D) Butterfly needles connected to a syringe. Photo: Dorothée Ordonneau.

6.1.1.1. Blood With Anticoagulant

For hematology, the blood sample must be collected with EDTA to preserve the size and shape of the cells. Heparin retards blood coagulation for up to eight hours, and its use is recommended for cytogenetic studies in tapirs. In addition, the field veterinarian should prepare a blood smear in the field (some laboratories recommend preparing blood smears without anticoagulant). It´s important to prepare the smear within a few hours of blood collection to avoid cell damage.

It is important to fill the tube with the specified volume of blood. If this is not done, the ratio of blood to anti-coagulant will not be correct and the cell count won't be accurate. The blood collected with anticoagulant should be homogenized soon after collection by slowly and continuously moving the container to mix the blood and the anticoagulant. The blood should then be refrigerated to reduce hemolysis. In the field it is often necessary keep the samples cool inside a cooler filled with ice. The sample must remain refrigerated until it is processed in the laboratory. For hematology, samples must be processed within 24 hours of collection.

6.1.1.2. Blood without Anticoagulant

For serum analyses for biochemistry and immunologic studies, samples must be collected without anticoagulant, and serum must be analyzed immediately or frozen and stored for later analysis. Samples must be refrigerated until processed in the laboratory. The processing of samples must occur within 24 hours. Glucose

determination should be performed within 3-4 hours of collection, otherwise the values become altered.

Samples are collected in vacuum tubes without anticoagulant, with or without gel. The amount of serum obtained per blood sample depends on the conditions of the animal, and is generally 50% or less. Hemolysis must be avoided, so the samples must be managed with care and protected from direct sunlight. After a short period of rest the blood should be refrigerated to reduce hemolysis. Again, in the field, it is often necessary to use a cooler filled with ice to refrigerate samples.

6.1.1.3. Handling and Storage of Blood

Once in the laboratory, a portion of the blood with anticoagulant should be used for hematology, and the remainder should be frozen for later analyses. Drops of blood without anticoagulant on filter paper should be stored in ambient temperature for later molecular analyses. The remainder of the sample should be centrifuged and its components, plasma, leukocytes and red blood cells, separated and frozen. Blood samples without anticoagulant should be also centrifuged and its components, serum and clot, separated and frozen.

It is necessary to centrifuge blood at 1500 rpm for 5-10 minutes. Aliquots of 1ml each can be stored in cryovials of 2ml and stored in a freezer or in liquid nitrogen. The ideal scenario is to store the cryovials in liquid nitrogen (-196ºC), followed by ultralow freezer (-86ºC), industrial freezer (-25ºC), and domestic freezer (-18ºC). It is important never to exceed half of the capacity of the cryovial; filling the cryovial beyond this may lead to it bursting open when put into liquid nitrogen.

6.1.1.4 Blood Smear

Blood smears are needed for differential leukocyte counts and the evaluation of blood parasites. To correctly prepare a blood smear, blood should be collected with or without anticoagulant from peripheral vessels, such as auricular veins, though if blood is maintained with anticoagulant for more than a few minutes, blood smears prepared from it can present staining artefacts or leukocyte aggregation. Collect the blood with a small syringe or heparinized capillary and place a small drop on a microscopy slide. Use another slide held at a 45° angle to spread the blood over the microscopy slide, then let the smear dry at ambient temperature, protected from insects and dust. Store the sample in a slide box at ambient temperature. Once in the laboratory, immerse the dried slide in absolute methanol, let it dry again, and then use the proper stains for microscopic evaluation. We recommend Giemsa stain. The interval between the preparation of the slide and its fixation with methanol should not exceed 4 hours, and the interval between fixation and staining should not exceed 2 weeks. Experts generally recommend that veterinarians prepare and stain at least two slides for each blood sample. Stained slides can be stored for several years at ambient temperature, protected from dust and direct sunlight.

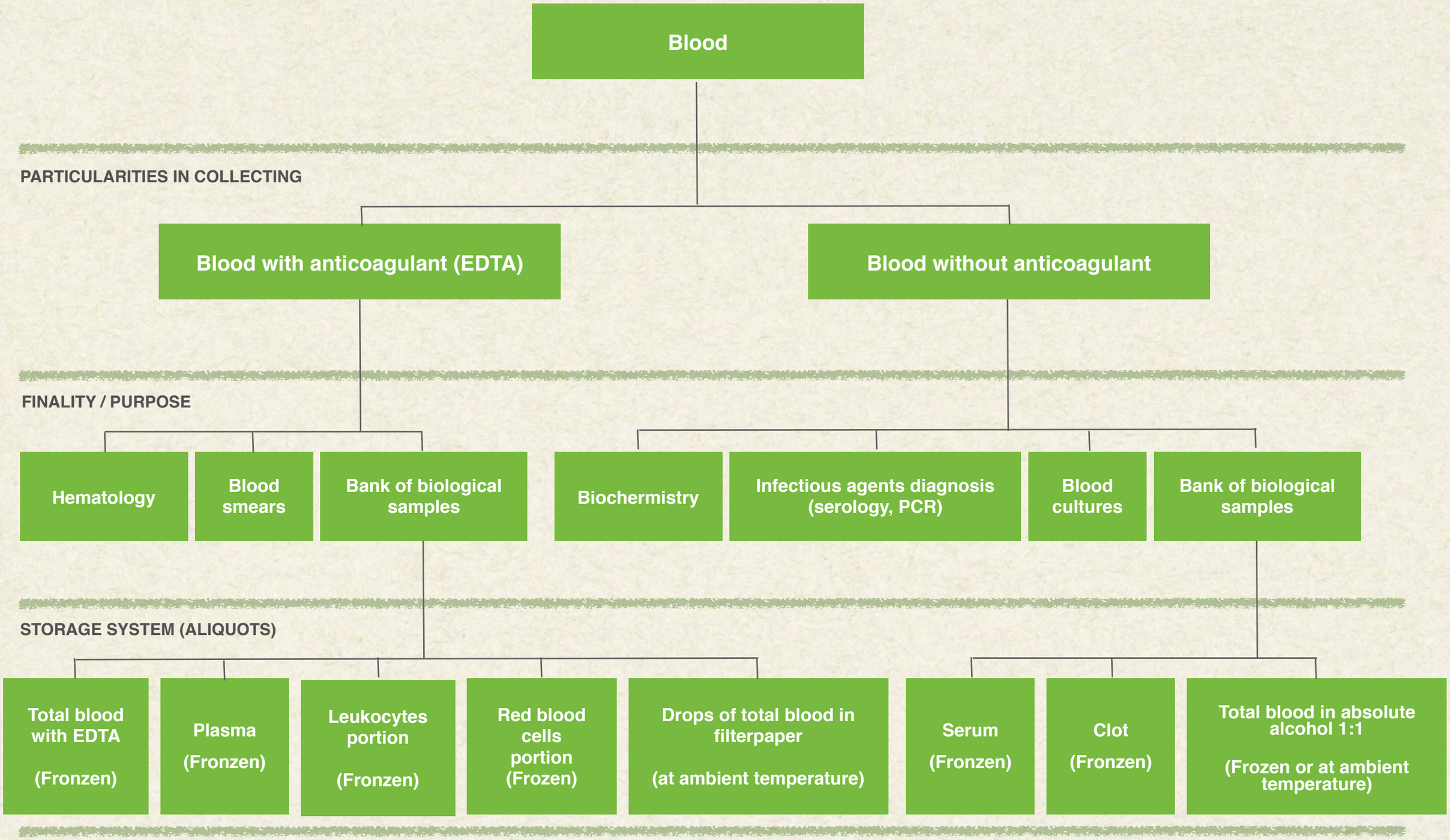


Figure 46 *- Suggested flowchart for blood samples. Picture Diagram: Renata Carolina Fernandes-Santos.*

6.1.2 Swabs for Microbiological Analysis

The collection of microbiological samples for bacterial cultures can be made with sterile swabs and proper nutritive/transport culture medium. Sampling techniques vary depending on the type of microorganism, being necessary to use swabs as transport medium in bacterial samples whereas fungi do not require that. A thorough aseptic process is required for collecting, processing and analyzing microbiological samples in order to avoid undesired contaminations and avoid accidental human infections. The use of sterile containers is strictly required.

Swabs of anatomical cavities (nasal, oral, ear, rectal, vaginal, urethra and preputial) are stored in transport media such as Stuart's swab. In the case of an abscess, the veterinarian should make an incision into the external layer after superficial disinfection, then drain the pus. Then he/she must collect the sample by rubbing the swab on the internal face of the external layer. Skin lesions and wounds should also be sampled. Swab samples must be refrigerated until transportation to the laboratory.

Hemocultures are recommended in cases where the occurrences of hematuria, hemoglobinuria, jaundice or septicemia are possible. Samplesare collected in 0.05-0.25% sodium polyanetosulfonate (SPS). Ammonium oxalate, sodium citrate and EDTA are not recommended because they inhibit some bacteria. The veterinary or technician who performs the sampling should follow the recommendations of the laboratory.

For the study of saprophyte fungi, the skin may be cleaned with ethanol 70%, and once dry, the sample is collected by rubbing the surface with a piece of sterile gauze. For pathogenic fungi in the skin, a sample is collected by scraping the periphery of the lesion with a blade and removing and collecting hairs from the affected area. In both cases, the samples are collected in sterilized bags, and stored without refrigeration in a dry, fresh and dark place until transportation to the laboratory.

Gallery 8 - Microbiological samples collection

Collection of microbiological samples for bacterial cultures using sterile swabs. Photos: Patrícia Medici and Renata Carolina Fernandes-Santos.

6.1.3 Fecal Samples

Fecal samples are used for the study of fecal parasites, hormones, diet and genetics. Whenever possible, feces should be obtained directly from the rectum. Here we describe sampling methodologies for the analysis of parasites and hormones.

6.1.3.1 Parasites: Fecal samples must be fresh. It is recommended to collect the sample from the central area of the

fecal pile in order to avoid environmental contamination. Samples must be kept refrigerated and processed within a maximum of 48 hours after collection.

Fecal samples for the analyses of fecal parasites should be stored in 5% formaldehyde solution (1 part of formaldehyde to 4 parts of fecal material; human fecal sample kits are effective) or refrigerated in 2.5% potassium dichromate solution (1:1) for subsequent analysis. There are two particularly successful field methods for processing fecal samples to analyze parasites: flotation and sedimentation. Neither of these methods can guarantee the identification of endoparasites (ova or larvae) down to species, but they permit researchers to identify them to the level of family. If identification of specific species is needed, it is necessary to consult with a veterinary specialized in parasites for methods of culturing ova and/or larvae in the field, and subsequent storage and handling.

Flotation Method: 3-5g of feces are placed in a small container (10-15ml) and mixed with a solution of a greater specific gravity than water, which will encourage the “flotation” of parasitic ova, cysts and some larvae. When commercial solutions are not available in the field, a super-saturated solution of sugar can be made by mixing table sugar and water. The container is filled with the mixture of feces and fecal flotation solution to form a positive meniscus and covered with a clean microscope slide. This is allowed to sit for 10-15 minutes, at which time the slide is removed. If ovum are present, they will have floated to the top and thus have “stuck”; they can then be covered by a coverglass and examined with a light microscope. In order to avoid structural deformations or even rupture of certain parasites ova due to the osmotic pressure in hypertonic medium is recommended to read the slide immediately after the “floating” process.

Centrifuge-flotation Method: When a centrifuge is available, the flotation technique in super-saturated solution of sucrose (specific density of 1.205 g/cm ^{3}) can provide better results in the identification of different parasite species. This technique results in a high concentration of eggs, cysts and oocysts on the surface of the sample, which helps detect parasites when infecting forms are eliminated in the feces in small amounts.

The supersaturated solution of sucrose (called Sheather’s solution) can be prepared by mixing three parts of an "A" solution (combine 128g of refined sugar with 100 ml of distilled water, then boil until the sugar is completely dissolved and the resulting solution is homogeneous and transparent) for each part of distilled water. The solution should be stored at 4ºC and should be used within 7 days of preparation; 0.5 to 1 ml of 37% formalin can be added if it is necessary to store the solution for longer periods of time.

The processing of the fecal samples using the centrifuge-flotation method consists of the following steps: (A) homogenizing 1 to 2 g of fecal sample in 11ml of super-saturated solution of sucrose; (B) filtering this material and putting it into tubes of 10 to 15ml; (C) centrifuging the tubes at 1,500RPM for 10 minutes; (D) collecting a drop of the supernatant with the aid of a platinum spatula; (G) putting this material on slide and (H) covering it with cover glass; (I) performing the reading of the blade using a bright-field microscope at 100 and 400x (modified by Fayer and Xiao 2008; Santos 2011).

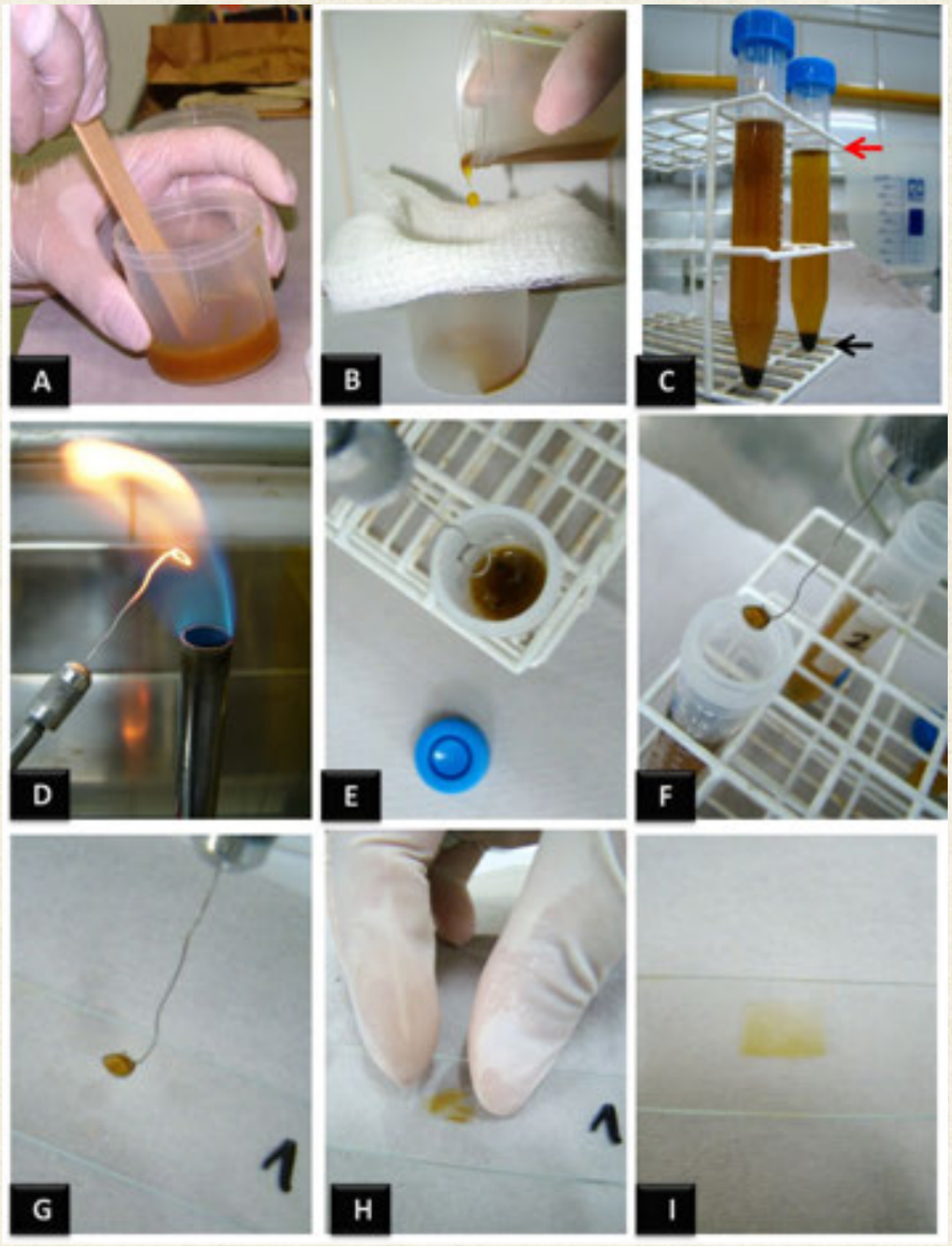


Figure 47 *- Photos: Renata Carolina Fernandes-Santos*

Sedimentation Method: This method allows the sedimentation of heavy parasitic ova that typically are not found with the flotation method (e.g. trematode eggs). To prepare samples using the sedimentation method, first 1 g of feces is thoroughly mixed with 5 ml of acetic acid. This is allowed to rest for 1 minute and then strained into a centrifuge tube. An identical volume of ether is added to this tube, mixed thoroughly and centrifuged for 1 minute at 400g (1,500 RPM, in general). The consequent sediment should contain the parasitic ova. The top most layers in the tube contain ether and acetic acid and should be appropriately discarded. The sediment should be mixed with a couple of drops of warm water and mixed thoroughly. This mixture is aspirated with a pipette and a couple of drops are placed on a clean microscope slide and examined with a light microscope.

6.1.3.2 Hormones: For the dosage of hormonal metabolites, samples should be frozen and sent to specialized laboratories. Fecal samples for hormonal analysis have to be as fresh as possible. It is recommended to homogenize the fecal sample and then collect an aliquot because the hormone metabolites are not equally distributed in the feces. Once the sample is collected it can be dried frozen or extracted in the field. The extracted sample can be stored until it is processed in an endocrinology laboratory.

6.1.4 Tissue Samples

See details about collection, handling and storage of tissue samples for genetics studies in the IUCN/SSC Tapir Specialist Group (TSG) Manual of Sampling Techniques for Genetic Analysis available online on the TSG Website (www.tapirs.org) in English, Spanish and Portuguese.

6.1.5 Hair

6.1.5.1 Genetics: See details about collection, handling and storage of hair samples for genetics studies in the IUCN/SSC Tapir Specialist Group (TSG) Manual of Sampling Techniques for Genetic Analysisavailable online on the TSG Website (www.tapirs.org) in English, Spanish and Portuguese.

6.1.5.2 Tricological Analyses: The hair should be collected preferably from the dorsal portion of the animal, carefully pulling both rough and thin hair manually. Hair samples should be transferred to a dry envelope or recipient, and kept away from humidity and excessive heat. If collected and stored properly, hair samples will remain intact for years.

6.1.6 Milk

If lactating females are captured, it is of interest to collect milk samples for bromatological analyses. For the case of females in captivity, collecting milk samples can be carried out under operant conditioning or anesthesia following this protocol as a guide (Fernandez y Quse, personal communication):

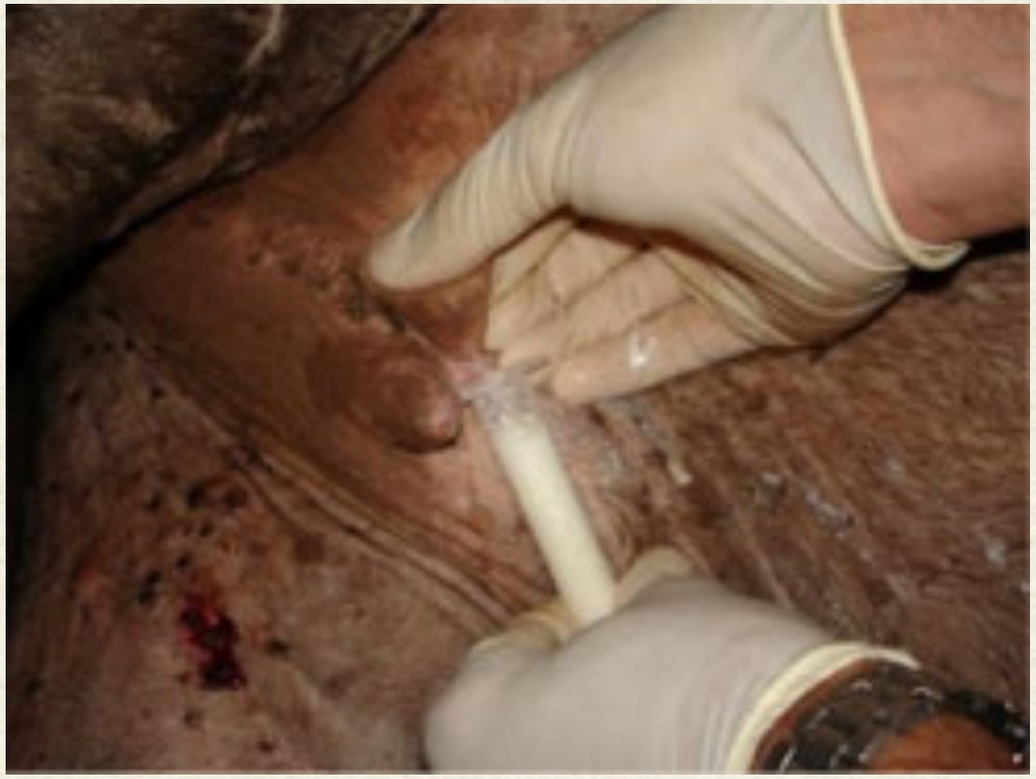


Figure 48 *- Collection of milk. Photo: Patrícia Medici.*

1. Try to position the tapir so that it is lying in lateral decubitus.

2. Clean the nipple with Chlorhexidine or warm water, and then, dry with a clean towel.

3. Massage the mamma to improve the slope of the milk.

4. Extract milk manually using latex gloves or properly cleaned hands.

5. Discard the first drops of milk obtained.

6. Collect remaining milk in sterile tubes or plastic flasks (like urinals) that can be hermetically sealed. The tubes or flasks should

be labeled with all pertinent information with indelible ink. A copy of this information should remain on site.

7. For bromatological analysis it is generally necessary to extract 5 to 10 milliliters of milk. 8. After extraction it is recommended to add a couple of drops of potassium dichromate 1% (bacteriostatic) to each 10 ml sample of milk.

9. The samples should be cooled and sent to the appropriate laboratory for analysis within the 24 hours of the extraction. The samples cannot be sent with ice, but they should be conditioned with dry ice and protected from light.

10. If the samples won't be sent immediately, they should be cooled in a freezer until the transfer. There must be a note explaining that the samples have been frozen.

11. If the samples are kept at at temperature of -20º C, they can be maintained for approximately 8 months.

6.1.7 Urine

The collection of urine by cystocentesis or urethral probing is not common in the field. Urine is usually only collected when the animal involuntarily urinates during the chemical restraint due to the relaxation caused by the anesthetic drugs. The urine should be collected in a sterile graduated screw capped flask, then kept under refrigeration until laboratorial analyses. Standard urinalysis and urine sediment analysis are recommended. Urine test dip stripscan be applied in the field for a rapid evaluation of possible metabolic/urinary diseases. A fraction of the

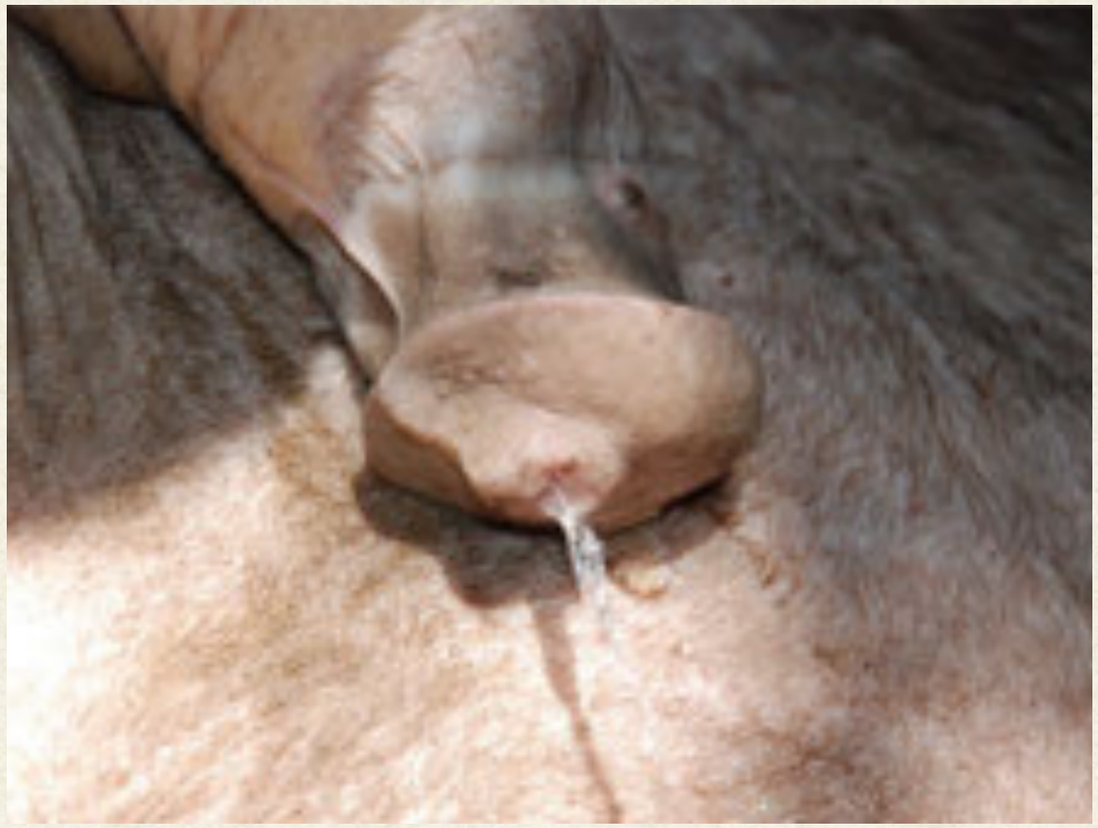


Figure 49 - *Spontaneous urination during anesthesia. Photo: Patrícia Medici.*

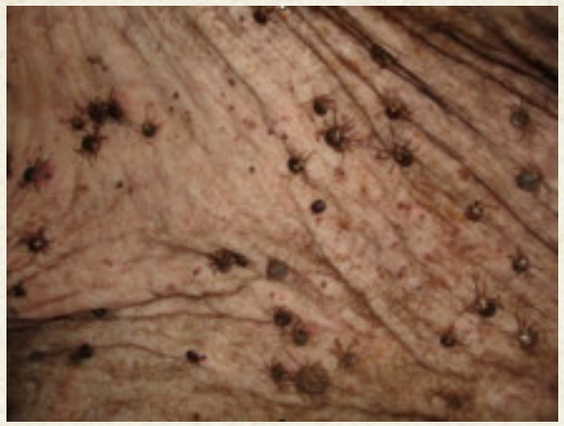
urine sample should be transferred to Eppendorf-like flasks or cryotubes for epidemiological analysis.

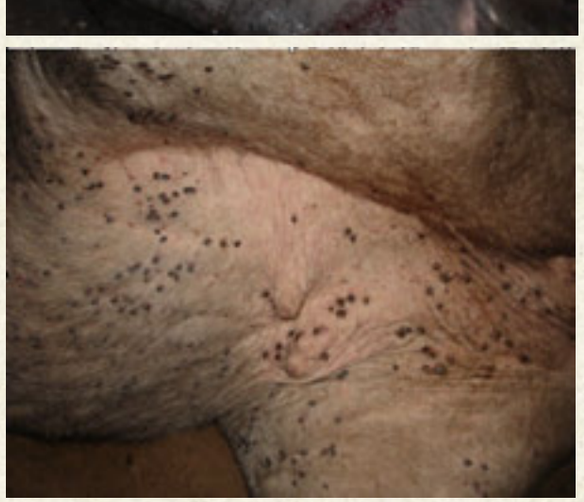
Urine samples can also be used for the diagnosis of Leptospirosis. For this purpose, the urine sample should be put in a saline solution (0.85%) in a proportion of 1:9; 0.5ml of this mixture should then be transferred to the appropriate culture medium.

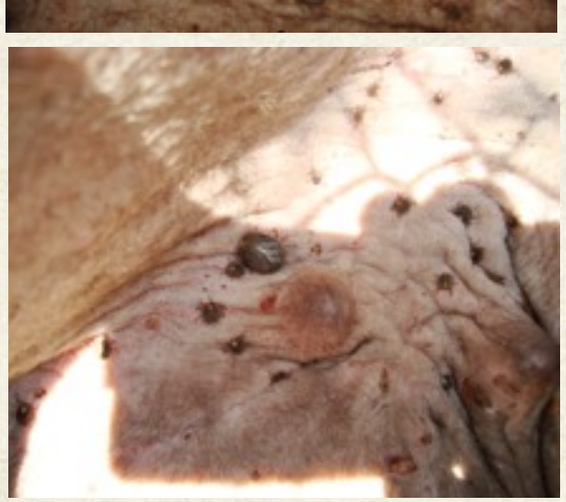
6.1.8 Ectoparasites

Ticks should be removed carefully, rotating them in order to avoid pulling off the bucal apparatus, which is critical for their microscopic identification. Laboratories recommend preserving ticks alive in small

Gallery 9 - Tick infestation







Tick infestation in wild lowland tapirs. Photos: Patrícia Medici.

containers with holes to allow for ventilation and a substrate that can be maintained humid. If it is necessary to store ticks for longer periods of time, samples should be preserved in ethanol 70%.

To determine if the interaction between parasite, wild host, and domestic host implies in epidemic risk, all the plethoric females may be gathered and submitted to a laboratory for larval cultures. To determine the parasitic load of an individual, all the plethoric females must be gathered and submitted to a laboratory for larval cultures. To determine the parasitic load of an individual, the common practice is to count all the ticks bigger than 4.5 mm diameter visible on one half of the body of the animal, and then double this number.

To sample the mites producing scabies, collect them from scrapings and hairs from the periphery of the affected area of the skin, and store them in sterile tubes with glycerin. Fleas can be collected directly from the body of the animal, and conserved in ethanol 70%.

6.1.9 Vaginal Cytology

Vaginal cytology is a tool used to assess the reproductive health of the females. Hygienize the vulva with Chlorhexidine or another disinfectant and insert a clean swab into the vagina (without touching the vulva), rotate the swab on the walls of the vagina, remove the swab and roll it over a microscope slide. It is best to then use alcohol to fixate the slide in the field then leave it to rest at ambient temperature and protected from insects and dust. In the laboratory, the Fast Panotic or Giemsa stains can be used for microscopic analysis.

6.1.10 Other Cytological Samples

Diagnostic clinical cytology (cythopathology) is an aid for the diagnostic clinical bacteriology, because it consists of the direct analysis of the liquid obtained from punctures and aspirations. It allows the identification

of the dominant cell type in a given inflammatory process, the status of the cells from the affected tissue, and in some cases the identification of the etiologic agent. It is generally a relatively simple technique that can be practiced in the field.

Table 5 *- Collection, handling and storage of biological samples in the field.*

Sample	Material	Collection Method	Handling	Storage
Non-Clotted Blood	flask with anticoagulant	venipuncture	homogenize and leave to rest	refrigeration
Clotted Blood	flask without anticoagulant	venipuncture	leave to rest	refrigeration
Blood Smears	microscopy slides	venipuncture	dry at ambient temperature	slide transport box, at ambient temperature
Skin/Tissue	jigger, scissors and flask	ear	100% alcohol	frozen, protected from light
Feces	flask	rectum	-	refrigeration
Urine	flask	spontaneous miction	-	refrigeration
Hair	flask or envelope	Manualpulling	-	ambient temperature
Milk	sterile flask	Manualmilking	-	refrigeration
Microbiological Sampling	sterile swab	nasal, oral, ear, rectal, vaginal, urethra and preputial cavities	nutritive/transport media	refrigeration
Vaginal Cytology	swab	rotation of the swab on the vaginal canal	microscopy slide, chemical fixation	slide transport box, at ambient temperature
Ectoparasites	perforated flask	manual collection, hand pulling	-	ambient temperature

RECOMMENDED LITERATURE

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Picture: Bill Konstant

7

Hematology and Blood Biochemistry

Hematology and Blood Biochemistry

Blood analyses provide information about the physiology and health status of the animal. These analyses allow the establishment of mean hematological and serum values and also the diagnosis of infections, anemia and nutritional deficiencies, hemoparasites and malfunctioning of internal organs. Basic hematology analyses can be carried out in the field with the assistance of a trained veterinarian and standardized techniques. Other analyses such as enzyme analyses, levels of glucose, lipids, cholesterol, vitamins and minerals are more difficult to carry out in the field, but serum samples can be collected, stored and later taken to a laboratory.

The blood analyses can be carried out by human laboratories, which will often be more accessible than veterinary clinical laboratories. It is important to ensure that cell counts are carried out manually and not by automatic equipment. In addition, make sure to get aquainted with the technique to be employed in the blood chemistry analyses; otherwise the results might be biased.

A complete hematological evaluation includes:red blood cell count (RBC), hemoglobin (Hb), packed cell volume (PCV), mean corpuscular volume (MCV), mean corpuscular hemoglobin (MCH), mean corpuscular hemoglobin concentration (MCHC), total white blood cell count (WBC) including differential and platelet counts, and the biochemical evaluation.

The red blood cell count can be done in a hemocytometer or Neubauer chamber by diluting blood in saline solution (NaCl 0,9%) or preferably Gower's solution (12.5 g sodium sulphate, 33.3 mL glacial acetic acid, q.s. 100 mL distilled water), in 1:200 proportion (10μL blood + 1990μL Gower's solution). Considering the chamber volume, the RBC result is the number of erythrocytes counted inside five of smaller central squares

multiplied by 10,000, and should be presented in 10^{12}cells/L of blood. For the total white blood cell count, Turk's solution (1 mL gentian violet solution, 3 mL glacial acetic acid, q.s. 100 mL distilled water) may be used in a 1:20 dilution (10μL blood + 190μL Turk solution). In this case, the result is the number of leukocytes counted inside the four larger corner squares multiplied by 50, and should be presented in 109 cells/L of blood (Figure X).

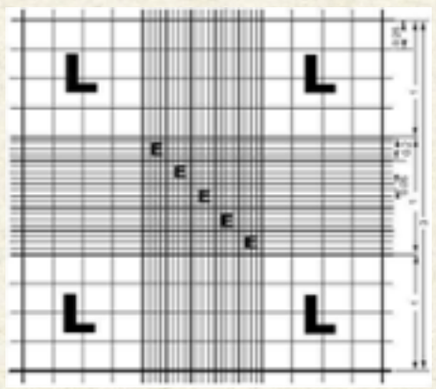

Figure 50 *- Neubauer Chamber.The central square is used for platelets and red cells. This square is split in 25 squares of width 0.2 mm (200µm). The four corner squares are use for the count of leucocytes.*

The interpretation of blood chemistry results should take into account the possible metabolic shifts caused by stress and restraint procedures, the clinical conditions of the animal at the moment of the chemical restraint, and the possible impacs of the blood collection and capture methods employed. For several parameters, the results represent only a snapshot of the biochemical condition of the blood at the moment of the capture, and cannot be interpreted as representative of the general physiological conditions of the animal. Indeed, when interpreting results, It is important to remember that capture-related stress and the use of anesthesia can both substantially modify some hematological and biochemical values.

Another strategy for better interpreting hematological exam results is to cross-reference them with the information available on the environment in which the tapir lives, the likely human interferences, and prior serological findings on tapirs and other species (including domestic animals) that might be in indirect contact with tapirs. Water pollution caused by human and/or domestic animal waste, farming pesticides, mining products and other pollutants may have cumulative effects both on the environment and on the tapirs, which can, in turn, affect biochemical and blood parameters in various ways.

The results of hematology and blood chemistry exams can be compared with the reference values developed for captive tapirs available in Physiological Data Reference Values for Tapir Species - International Species Information System (ISIS) published in 2006: www.isis.org

Table 6 *- Hematological and biochemistry parameters that should be evaluated and Standard International (SI) Unit for each parameter.*

Hematological parameter	SI Unit
Red blood cell count	10^{12}/L
Hemoglobin	g/dL
Packed cell volume	L/L
Reticulocytes	%
MCV	fL
MCH	pg
MCHC	g/dL
White blood cell count	10^{9}/L
Eosinophils	10^{9}/L
Basophils	10^{9}/L
Lymphocytes	10^{9}/L
Monocytes	10^{9}/L
Band neutrophils	10^{9}/L
Segmented neutrophils	10^{9}/L
Total neutrophils	10^{9}/L
Platelet count	10^{9}/L
Alanine Aminotransferase (ALT)	U/L
Aspartate Aminotransferase (AST)	U/L
Gamma Glutamyl Transferase (GGT)	U/L

Biochemistry parameter	SI Unit
Urea Nitrogen (BUN)	mmol/L
Uric Acid	µmol/L
Creatinine	µmol/L
Creatinine Phosphokinase (CPK)	U/L
Alkaline Phosphatase (ALP)	U/L
Glucose	mmol/L
Total Cholesterol	mmol/L
HDL Cholesterol	mmol/L
LDL Cholesterol	mmol/L
VLDL Cholesterol	mmol/L
Triglyceride	mmol/L
Fibrinogen	µmol/L
Total Protein	g/L
Albumin	g/L
Globulin	g/L
Albumin/Globulin	Alb/Glob
Cholinesterase	U/L
Amylase	U/L
Total Bilirubin	µmol/L

Biochemistry parameter	SI Unit
Direct Bilirubin	µmol/L
Indirect Bilirubin	µmol/L
Magnesium	mmol/L
Sodium	mmol/L
Potassium	mmol/L
Calcium	mmol/L
Phosphorus	mmol/L
Chloride	mmol/L
Iron	µmol/L
Cortisol	nMol/L
Tetosterone	nMol/L
Progesterone	nMol/L
Estrogen	nMol/L
Lactate dehydrogenase (LDH)	U/L

LOWLAND TAPIR CONSERVATION INITIATIVE, Brazil

Reference values for HEMATOLOGICAL and BIOCHEMICAL PARAMETERS of wild lowland tapir *Tapirus terrestris* (Medici, Mangini & Fernandes-Santos 2014)

HEMATOLOGICAL PARAMETERS	SI Unit	ATLANTIC FOREST (AF)										PANTANAL (PA)										AF + PA									
		N	Mean	Min	Q1	Median	Q3	Max	SD	SE		N	Mean	Min	Q1	Median	Q3	Max	SD	SE		N	Mean	Min	Q1	Median	Q3	Max	SD	SE	
Red blood cell count	10^{12}/L	22	4.56	2.68	3.32	4.59	5.35	6.74	1.11	0.24	1,2	40	5.95	2.71	4.78	5.67	6.65	11.58	1.78	0.28	1,2	62	5.46	2.68	4.52	5.44	6.39	11.58	1.70	0.22	1
Hemoglobin	g/dL	22	9.09	7.10	8.40	9.15	9.70	10.40	0.97	0.21	1,2	21	10.96	7.70	10.50	11.00	11.50	13.30	1.48	0.32	1,2	43	10.00	7.10	9.00	9.70	11.00	13.30	1.55	0.24	1
Packed cell volume	L/L	20	0.28	0.25	0.26	0.28	0.30	0.33	0.02	0.01	1,2	39	0.34	0.26	0.31	0.34	0.37	0.44	0.05	0.01	1,2	59	0.32	0.25	0.28	0.32	0.35	0.44	0.05	0.01	1
MCV	fL	22	62.36	48.00	55.00	57.00	78.00	88.00	13.03	2.78	1	28	57.99	31.00	50.55	57.25	64.00	89.70	12.37	2.34	1	50	59.91	31.00	51.50	57.25	65.10	89.70	12.73	1.80	1
MCH	pg	22	20.41	15.00	18.00	19.00	25.00	30.00	4.59	0.98	1	20	20.16	16.00	17.65	19.65	22.00	27.10	2.98	0.67	1	42	20.29	15.00	18.00	19.40	23.20	30.00	3.87	0.60	1
MCHC	g/dL	21	32.81	31.00	32.00	33.00	33.00	34.00	0.84	0.18	1	21	32.89	27.70	32.60	33.30	34.10	35.00	1.87	0.41	1	42	32.85	27.70	32.00	33.00	33.70	35.00	1.43	0.22	1
White blood cell count	10^3/L	22	8.87	6.90	7.70	8.35	9.60	13.30	1.67	0.36	1	36	10.21	5.16	7.69	10.40	12.63	15.15	2.87	0.48	#	58	9.70	5.16	7.70	9.60	11.45	15.15	2.55	0.34	#
Eosinophils	10^3/L	22	0.59	0.00	0.08	0.18	0.56	2.78	0.86	0.18	#	30	0.42	0.00	0.00	0.08	0.64	2.61	0.65	0.12	#	52	0.49	0.00	0.00	0.15	0.60	2.78	0.74	0.10	#
Basophils	10^3/L	22	0.03	0.00	0.00	0.00	0.08	0.13	0.04	0.01	1	20	0.00	0.00	0.00	0.00	0.00	0.05	0.02	0.00	1	42	0.02	0.00	0.00	0.00	0.00	0.13	0.04	0.01	1
Lymphocytes	10^3/L	22	2.21	0.81	1.62	2.16	2.55	4.01	0.82	0.17	1	31	2.75	0.32	1.54	2.41	3.81	6.43	1.50	0.27	#	53	2.53	0.32	1.60	2.29	3.07	6.43	1.28	0.18	1
Monocytes	10^3/L	22	0.12	0.00	0.08	0.11	0.14	0.36	0.08	0.02	1,2	31	0.37	0.00	0.10	0.25	0.44	1.71	0.41	0.07	#,2	53	0.26	0.00	0.08	0.15	0.27	1.71	0.34	0.05	#
Band neutrophils	10^3/L	22	0.31	0.00	0.16	0.26	0.49	0.83	0.21	0.05	1,2	31	0.20	0.00	0.00	0.05	0.25	1.28	0.32	0.06	1,2	53	0.24	0.00	0.00	0.16	0.36	1.28	0.29	0.04	1
Segmented neutrophils	10^3/L	22	5.60	2.97	4.72	5.30	6.46	8.81	1.39	0.30	*	31	5.65	0.18	3.53	5.27	8.07	11.84	2.96	0.53	*	53	5.63	0.18	4.26	5.27	7.12	11.84	2.41	0.33	*
Total neutrophils	10^3/L	18	5.83	4.40	5.19	5.48	6.32	8.89	1.05	0.25	#	29	6.11	0.18	3.60	5.40	8.24	15.68	3.38	0.63	#	47	6.00	0.18	4.40	5.44	7.18	15.68	2.72	0.40	#
Platelet count	10^3/L	15	297.40	148.00	255.00	310.00	352.00	398.00	70.00	18.07	1,2	15	234.93	40.00	186.00	242.00	278.00	354.00	81.56	21.06	#,2	30	266.17	40.00	209.00	275.50	331.00	398.00	81.15	14.82	1

BIOCHEMICAL PARAMETERS	SI Unit	ATLANTIC FOREST (AF)										PANTANAL (PA)										AF + PA									
		N	Mean	Min	Q1	Median	Q3	Max	SD	SE		N	Mean	Min	Q1	Median	Q3	Max	SD	SE		N	Mean	Min	Q1	Median	Q3	Max	SD	SE	
Alanine Aminotransferase (ALT)	U/L	20	9.88	5.00	7.00	10.00	12.00	15.00	2.91	0.65	#,2	38	19.13	12.00	16.00	18.00	22.00	31.00	4.36	0.71	1,2	58	15.94	5.00	12.00	15.50	20.00	31.00	5.90	0.78	1
Aspartate Aminotransferase (AST)	U/L	21	62.51	39.00	52.00	60.00	77.00	87.00	14.99	3.27	1,2	40	72.03	49.00	61.50	67.50	78.00	115.00	15.83	2.50	#,2	61	68.75	39.00	60.00	67.00	77.00	115.00	16.08	2.06	1
Gamma Glutamyl Transferase (GGT)	U/L	20	15.89	4.10	6.10	9.90	22.40	57.00	14.45	3.23	1,2	39	15.77	9.00	13.00	16.00	18.00	23.00	3.53	0.56	1,2	59	15.81	4.10	11.00	15.00	18.00	57.00	8.75	1.14	1
Blood Urea Nitrogen (BUN)	mmol/L	29	5.89	2.86	5.00	5.71	7.14	11.07	2.02	0.37	1	44	5.14	2.86	3.93	5.00	5.71	12.50	1.82	0.27	1	73	5.44	2.86	3.93	5.36	6.43	12.50	1.92	0.23	1
Uric Acid	µmol/L	7	30.59	5.95	11.90	17.84	59.48	65.43	24.63	9.31	#	40	14.72	5.95	11.90	11.90	17.84	29.74	6.17	0.98	1	47	17.08	5.95	11.90	11.90	17.84	65.43	12.00	1.75	1
Creatinine	µmol/L	21	60.20	35.36	44.20	53.04	70.72	106.08	18.25	3.98	1,2	41	105.43	61.88	97.24	106.08	114.92	141.44	18.37	2.87	1,2	62	90.11	35.36	61.88	97.24	106.08	141.44	28.22	3.58	1
Creatinine Phosphokinase (CPK)	U/L	14	450.93	60.00	103.00	198.50	646.00	1526.00	498.80	133.31	*,2	40	170.00	56.00	100.50	126.50	162.00	772.00	146.11	23.10	*,2	54	242.83	56.00	101.00	131.50	210.00	1526.00	303.61	41.32	*
Alkaline Phosphatase (ALP)	U/L	18	24.44	10.00	15.00	21.50	31.00	47.00	11.79	2.78	1,2	39	13.49	2.00	9.00	13.00	18.00	29.00	5.91	0.95	1,2	57	16.95	2.00	11.00	14.00	20.00	47.00	9.61	1.27	1
Glucose	mmol/L	13	6.93	3.05	4.61	6.72	9.05	10.71	2.43	0.67	#	40	6.04	3.66	4.94	5.80	6.99	9.38	1.54	0.24	#	53	6.26	3.05	4.88	5.88	7.27	10.71	1.82	0.25	1
Total Cholesterol	mmol/L	22	3.52	2.38	2.90	3.38	3.99	5.31	0.79	0.17	1	39	3.35	2.25	2.77	3.34	3.83	4.74	0.69	0.11	1	61	3.41	2.25	2.89	3.34	3.83	5.31	0.72	0.09	1
HDL Cholesterol	mmol/L	6	1.64	1.14	1.24	1.55	2.05	2.28	0.45	0.18	*,2	41	2.17	1.30	1.71	2.05	2.56	3.26	0.57	0.09	*,2	47	2.11	1.14	1.55	2.05	2.54	3.26	0.58	0.09	*
LDL Cholesterol	mmol/L	2	2.60	2.28	2.28	2.60	2.93	2.93	0.46	0.32	*,2	39	0.93	0.39	0.62	0.88	1.17	1.79	0.36	0.06	*,2	41	1.01	0.39	0.67	0.88	1.17	2.93	0.51	0.08	*
VLDL Cholesterol	mmol/L	2	0.13	0.10	0.10	0.13	0.16	0.16	0.04	0.03	*	41	0.23	0.05	0.10	0.23	0.34	0.54	0.13	0.02	*	43	0.23	0.05	0.10	0.23	0.34	0.54	0.13	0.02	*
Triglyceride	mmol/L	14	0.54	0.20	0.35	0.45	0.70	1.03	0.27	0.07	#	40	0.51	0.12	0.22	0.49	0.76	1.20	0.30	0.05	#	54	0.52	0.12	0.28	0.49	0.73	1.20	0.29	0.04	#
Fibrinogen	µmol/L	5	7.03	5.73	5.73	6.17	7.20	10.29	1.92	0.86	#	*	*	*	*	*	*	*	*	*	*	5	7.03	5.73	5.73	6.17	7.20	10.29	1.92	0.86	#
Total Protein	g/L	22	75.97	53.00	65.20	77.00	87.00	99.00	13.70	2.92	1,2	40	63.55	56.00	61.00	63.00	66.50	72.00	4.25	0.67	1,2	62	67.96	53.00	61.00	65.00	70.30	99.00	10.58	1.34	#
Albumin	g/L	14	23.93	21.00	22.00	24.00	26.00	27.00	2.06	0.55	1,2	40	16.08	11.00	13.00	16.00	19.00	21.00	3.10	0.49	1,2	54	18.11	11.00	14.00	18.00	21.00	27.00	4.49	0.61	1
Globulin	g/L	14	57.14	32.00	54.00	57.50	65.00	76.00	12.45	3.33	1,2	39	47.28	41.00	45.00	47.00	50.00	56.00	3.91	0.63	1,2	53	49.89	32.00	45.00	48.00	53.00	76.00	8.32	1.14	1
Albumin/Globulin	Alb/Glob	4	0.43	0.30	0.35	0.45	0.50	0.50	0.10	0.05	*	42	0.32	0.20	0.20	0.30	0.40	0.50	0.11	0.02	*	46	0.33	0.20	0.20	0.30	0.40	0.50	0.11	0.02	*
Cholinesterase	U/L	14	168.84	0.70	0.90	1.30	378.00	609.00	220.75	59.00	*	40	262.18	120.00	189.50	233.00	299.00	540.00	113.82	18.00	*	54	237.98	0.70	139.00	228.00	312.00	609.00	152.28	20.72	*
Amylase	U/L	4	283.42	242.54	250.44	280.74	313.71	329.67	42.25	21.12	1	*	*	*	*	*	*	*	*	*	*	4	283.42	242.54	250.44	280.74	313.71	329.67	42.25	21.12	1
Total Bilirubin	µmol/L	19	12.60	6.84	8.55	11.97	15.39	27.36	5.41	1.24	#,2	39	4.93	2.05	3.59	4.79	6.16	8.55	1.75	0.28	1,2	58	7.44	2.05	4.62	6.07	8.55	27.36	4.95	0.65	1
Direct Bilirubin	µmol/L	19	3.51	0.00	1.71	3.42	5.13	6.84	1.66	0.38	#,2	41	0.86	0.17	0.34	0.68	1.54	1.88	0.60	0.09	1,2	60	1.70	0.00	0.51	1.37	1.88	6.84	1.62	0.21	1
Indirect Bilirubin	µmol/L	18	7.79	3.42	5.13	7.70	10.26	11.97	2.50	0.59	#,2	40	3.95	1.54	2.65	3.85	5.13	6.67	1.46	0.23	1,2	58	5.14	1.54	3.25	5.04	6.67	11.97	2.56	0.34	1
Magnesium	mmol/L	21	0.65	0.16	0.49	0.66	0.74	1.44	0.32	0.07	1,2	40	0.69	0.45	0.58	0.66	0.78	0.99	0.14	0.02	1,2	61	0.68	0.16	0.58	0.66	0.78	1.44	0.22	0.03	1
Sodium	mmol/L	21	132.81	110.00	129.00	133.00	138.00	155.00	9.28	2.03	#	42	135.00	128.00	130.00	134.00	139.00	148.00	5.45	0.84	#	63	134.27	110.00	130.00	134.00	139.00	155.00	6.97	0.88	#
Potassium	mmol/L	21	3.75	2.50	3.10	3.50	4.10	5.30	0.81	0.18	#	39	3.63	2.70	3.10	3.40	4.10	4.80	0.65	0.10	#	60	3.67	2.50	3.10	3.45	4.10	5.30	0.71	0.09	#
Calcium	mmol/L	21	2.30	1.78	2.03	2.23	2.50	3.28	0.38	0.08	1,2	39	2.42	2.05	2.28	2.43	2.55	2.75	0.20	0.03	1,2	60	2.38	1.78	2.23	2.38	2.55	3.28	0.28	0.04	1
Phosphorus	mmol/L	16	0.89	0.45	0.63	0.72	1.20	1.62	0.36	0.09	1,2	39	1.04	0.58	0.81	1.00	1.26	1.74	0.29	0.05	1,2	55	1.00	0.45	0.74	0.97	1.23	1.74	0.32	0.04	1
Chloride	mmol/L	13	108.34	95.80	104.00	110.00	113.80	118.00	6.91	1.92	1,2	40	99.45	90.00	94.00	97.50	103.50	114.00	6.61	1.04	#,2	53	101.63	90.00	95.00	101.00	108.00	118.00	7.66	1.05	1
Iron	µmol/L	13	15.17	8.23	10.02	13.78	15.75	30.43	7.22	2.00	1,2	40	13.75	8.06	11.19	13.96	15.57	21.84	3.69	0.58	1,2	53	14.10	8.06	10.92	13.78	15.57	30.43	4.76	0.65	1

[1] indicates parameters that were significantly different ($p<0.05$) from ISIS (Teare 2006), "all ages all classes" category.
[2] indicates parameters that were significantly different ($p<0.05$) when comparing AF and PA data.
# indicates parameters that were not significantly different ($p<0.05$) from ISIS (Teare 2006).
* indicates parameters absent from ISIS (Teare 2006) or from PA data.

MCV = Mean Corpuscular Volume; MCH = Mean Corpuscular Hemoglobin; MCHC = Mean Corpuscular Hemoglobin Concentration; HDL = High Density Lipoprotein; LDL = Low Density Lipoprotein; VLDL = Very Low Density Lipoprotein
N = valid sample size; Q1 = lower quartile; Q3 = upper quartile; SD = standard deviation; SE = standard error

Medici EP, Mangini PR, Fernandes -Santos RC. 2014. Health Assessment of Wild Lowland Tapir (*Tapirus terrestris*) Populations in the Atlantic Forest and Pantanal Biomes(1996-2012), Brazil. Journal of Wildlife Diseases, 50(4):817-828.

RECOMMENDED LITERATURE

Medici EP; Mangini PR; Fernandes-Santos RC. 2014. Health Assessment of Wild Lowland Tapir (Tapirus terrestris) Populations in the Atlantic Forest and Pantanal Biomes, Brazil (1996-2012). In: Journal of Wildlife Diseases 50(4):817-828.

Teare JA. 2006. Physiological Data Reference Values for Tapir Species. International Species Information System IUCN/SSC Tapir Specialist Group Veterinary Committee.

Picture: Luciano Candisani

8

Diagnostic for Selected Infectious Agents

Diagnostic for Selected Infectious Agents

The increased fragmentation of tapir habitat and the subsequent rise in contact between tapirs and domestic animals have been flagged as potential causes of the emergence of infectious diseases in tapir populations (Mangini et al. 2012). In populations where frequent interactions between wild animals and domestic livestock exist, mutual transmission of pathogens can occur, and may even affect human populations.

Due to the potential implications for conservation and human health, several field researchers have been investigating the impact of infectious diseases on tapirs. As mentioned in the introduction, available field information on tapir health is scarce and most of it comes from immunological investigation and is commonly based in serological screening. These studies can help with the identification of the role played by wildlife species in some diseases, and provide an important scientific baseline for the implementation of control measures if an epidemic disease arises. However, positive results in serologic tests do not inherently entail disease occurrence. Frequently previous exposure to an infectious agent can produce positive results. Furthermore, an important limitation of serological tests in wild animals is the lack of pattern antibodies and specific antigens, which limits the applicability of the techniques and the accuracy of the results (Mangini et al. 2012). Thus, it is quite difficult to use serologic tests alone to test hypotheses related to disease transmission between domestic animals and tapirs.

To ensure research results are useful and applicable, the infectious agents to investigate should be based on previous knowledge about the diseases that could be expected to occur in the study area according to the local prevalence and diversity of domestic animals nearby (especially

ungulates). Local governmental agencies and other epidemiology and sanitary agencies, and human and animal health organizations can offer additional guidance and provide information to improve the interpretation of research results. It is also always recommended to compare the serological results with any and all serological information for other local species, especially domestic livestock and humans, in order to understand the importance of the tapir in the epidemiological chain.

The decision to screen for diseases of mandatory reporting (either to the OIE - World Organization for Animal Health - or local agencies) should be carefully considered by the veterinarian in charge, and made only after taking into account the potential economic and social consequences. If a researcher decides to screen for such diseases, during the interpretation of results and their true significance on health, it is particularly important to consider the sensibility and specificity of the laboratory techniques, and the characteristics of the infectious agent and of the tapir species. Again, in some cases, a positive result does not automtically entail a health crisis. For instance, a PCR for herpesvirus will always be positive, but the impact of HV infection depends of many other factors that could exacerbate or nullify the implications of the positive result (Benoit Thoisy personal communication).

In order to more comprehensively understand the role of etiologic agents in tapir populations, serological results should be accompanied by alternative and complementary diagnostic methods. Physical examinations are one of the most relevant assessments of the health of an animal and, consequently, of its population. Clinical findings must be compiled and evaluated in conjunction with laboratory exams (hematology, biochemistry, parasitology, urinalysis, microbiology). Positive results in serological screening reactions should be submitted for confirmatory tests (usually with higher diagnostic specificity, though this comes with a higher cost and lower processing speed), like PCR or isolation of a specific agent (Richtzenhain & Soares 2007).

Common and important topic in the One Health and Conservation Medicine approaches are zoonosis. Climate change is also a big issue in wildlife medicine, especially in tropical countries it may have implcations for the prevalence of vector-borne diseases. One of the most important zoonotic diseases is Tuberculosis (TB), caused by some species of the genusMycobacterium such as Mycobacterium bovis and M. pinnipedii. Both belong to the Mycobacterium tuberculosis complex (MTC). M. pinnipedii is a mycobacteria from pinnipeds that can be transmitted to tapirs (Tapirus indicus and T. terrestris) in captivity; it was discovered in the last decade by professionals from Australia and Argentina (Cousins et al. 2003; Bastida et al. 2010; Bastida et al. 2011; Hoyer 2011).

Table 7 *- Suggested infectious agents relevant for surveys in tapirs.*

Agents Category	Infectious Agent / Infectious Disease
Viral	Vesicular Stomatitis Virus / Vesicular Stomatitis
	Bluetongue Virus / Bluetongue
	Infectious Bovine Rhinotracheitis (IBR) Virus / Infectious Bovine Rhinotracheitis (IBR)
	Foot and Mouth Disease Virus / Foot and Mouth Disease
	Equine Herpesvirus
	Equine Influenza Virus / Equine Influenza
	Eastern Equine Encephalomyelitis (EEE) Virus / Eastern Equine Encephalitis (EEE)
	Western Equine Encephalomyelitis (WEE) Virus / Western Equine Encephalitis (WEE)
	Venezuelan Equine Encephalitis (VEE) Virus / Venezuelan Equine Encephalitis (VEE)
	Rabies Virus/ Rabies
	Equine Rhinovirus
	Bovine Viral Diarrhea (BVD) Virus / Bovine Viral Diarrhea (BVD)
	Bovine Leukemia Virus / Enzootic Bovine Leucosis (EBL)
	Pseudorabies Virus (Suid Herpesvirus type 1) / Aujeszky's Disease
	Porcine Parvovirus
	Parainfluenza Virus / Parainfluenza 3
	Equine Infectious Anemia (EIA) Virus / Equine Infectious Anemia (EIA)
	West Nile Virus
Protozoans	*Trypanosoma*spp. / Chagas Disease
	Leishmania spp. / Leishmaniosis
	Babesia spp. / Babesiosis
	Toxoplasma gondii / Toxoplasmosis
Bacterial	*Brucella* spp. / Brucelosis
	Salmonella spp. / Salmonelosis
	Mycobacterium bovis; *M.tuberculosis*; *M. pinnipedii*; *M. avium* / Tuberculosis (TB) *M. paratuberculosis*/ Johne's Disease or Paratuberculosis
	Chlamydophyla spp. / Chlamydiosis
	Leptospira interrogans / Leptospirosis *Rickettsia* spp. / Rickettsiosis *Erlichia* spp. / Erlichiosis *Anaplasma* spp. / Anaplasmosis *Clostridium tetani* / Tetanus

Table 8 *- List of Leptospira interrogans serovars.*

Pomona	*Hebdomadis*	*Autumnalis*	*Tarassovi*
Hardjo	*Copenhageni*	*Castellonis*	*Mini*
Icterohaemorrhagiae	*Javanica*	*Bataviae*	*Guaicurus*
Grippotyphosa	*Panama*	*Butembo*	*Ballum*
Canicola	*Pyrogenes*	*Whitcombi*	*Sejroe*
Bratislava	*Wolffi*	*Cynopteri*	*Szwajizak*
Andamana	*Shermani*	*Sentot*	*Saxkoebing*
Australis	*Patoc*		

Table 9 *- Categorization of disease relevance for tapir population viability and conservation (Medici et al 2007, 2008; Mangini et al 2012).*

Relevance	Clinical diseases reported for tapirs	Diseases with serological evidence	Possible diseases
High	Foot-and-Mouth Disease Campylobacteriosis Tuberculosis	Equine Infectious Anemia Equine Encephalitis (WEE; EEE) Vesicular Stomatitis Leptospirosis Trypanossomosis	Brucellosis Intoxication (pesticides, heavy metals) Rabies
Medium	Balantidiosis Giardiosis Salmonellosis	Babesiosis Encephalomyocarditis Virus Infectious Bovine Rhinotracheitis	Pseudorabies (Aujeszky's Disease) Clostridiosis Leishmaniasis
Low	Blepharitis Respiratory Problems Tetanus Hemochromatosis	Equine Herpesvirus Bluetongue Mycoplasmosis Toxoplasmosis Enteric Red-and-Mouth Disease	Bovine Viral Diarrhea Influenza Swine Parvovirosis Rhinoviruses
Null	Actinomycosis Keratitis Diabetes Vesicular Exanthematous Disease Filariasis Laminitis Phleas Scabies Schistosomiosis		

8.1.1. Bacterial Diseases

The most common bacterial infections in tapirs include enteritis, tuberculosis, leptospirosis and tetanus. All are known to cause relevant clinical problems in captive animals. Septicemia and enteritis caused by Salmonella sp., Streptococcus sp. or Pasteurella sp. are also frequently reported in tapirs (Cubas 1996; Janssenet al. 1999; Mangini et al. 2002; Janssen 2003; Mangini et al. 2012).

1. **Salmonella sp.** Salmonellosis assumes one of the following forms: peracute septicemia, acute enteritis, chronic enteritis or a subclinical state. This disease has been reported in tapirs in captivity. Salmonella tiphimurium was associated with fatal septicemia in lowland tapirs, and S. pomona was isolated from a neonatal Baird's tapir with acute gastrointestinal distress. The occurrence of Salmonellosis in zoos generally coincides with the rainy season. Diagnostic tests for Salmonella can be carried out as a routine bacterial culture on an enteric medium such as Selenite medium or hectone enteric agar. In general the treatment consists of antibiotics (susceptibility testing) and fluid therapy (Ramsay & Zainuddin 1993).

2. **Mycobacterium sp.** Mycobacteria sporadically affects captive tapirs (Janssen et al. 1996). Mycobacterium bovis is the species most frequently diagnosed in tapirs in captivity, but tapirs have also been described to be susceptible to M. tuberculosis and M. avium. Myobacterial infections primairly affect the lungs and mediastinal lymph nodes. As mentioned previously, in the last decade veterinarians began to diagnose Tuberculosis (TB) in tapirs in captivity that is produced by a new species of mycobacteria belonging to Mycobacterium tuberculosis complex (MTC): Mycobacterium pinnipedii (Cousins et al. 2003). This species is transmitted TB through a primary host of marine origin and is considered aggressive and highly contagious, not only to humans but also to other wild and domestic species. For instance, M. pinipedii has affected not only a variety of species of wild mammals in captivity (tapirs, camels, llamas, gorilas, etc.) kept in zoos and aquariums near pinnipeds affected withTB, but keepers and technical staff working with or in close contact with these marine mammals have also contracted the disease. The new data on the transmission of M. pinipedii has led Dr. Ricardo Bastida to hypothesize that the origin of Pre-European TB in South America was caused by the frequent contact that resident hunter-gatherers had with pinnipeds as a food source for thousands of years during the years before colonization. After TB affected those hunter-gatherers that consumed pinnipeds, it was then transmitted to other organized cultural societies in South America through direct contact with the infected parties (Bastida et al.2010; Bastida et al. 2011).

M. pinnipedii has affected approximately 17 species of terrestrial and aquatic mammals, both in the wild and in captivity. This includes several reports of the strain of TB in Tapirus terrestris and Tapirus indicus in different zoos around the world (Bastida et al. 1999; Bastida et al. 2010; Bastida et al. 2011; Jurczynski et al. 2011; Hoyer 2011).

There is currently no specific and efficient test to diagnose an M. pinnipedii infection. Only a combination of different tests can be used to make an accurate diagnosis. The first step in this process is a comparative intradermal skin test (applied 0.1ml of BPPD in the skin inguinal region), combined with serological exams (ELISA, or Chembio DPP Vet TB Assay for elephants). These serological exams are not made for tapirs, and some years of test results are still needed to

properly evaluate the accuracy of the results for tapirs. At the very least, it is known that due to their way of life, tapirs are often in contact with non pathogen mycobacteria (bedding material, ground exploration), which can lead to false positives. Thus if the serological exam results are positive or unclear, further exams have to be done to properly diagnose or rule out TB. Some of these tests include: thoracic X-ray, pulmonary or tracheal and gastric wash and cultures or molecular techniques. In captivity, veterinarians should test each tapir a minimum of once a year, before any transfer, or if tapirs show possible symptoms of TB infection.

We currently have little information on the prevalence of M. pinnipedii in wild tapirs and whether it has a significant effect on free-ranging populations. With the design of new, less-invasive testing methods for Mycobacterium (DNA-based testing, ELISA, BTB tests etc.), the TSG Veterinary Committee encourages individuals working with free-ranging tapirs to use these methods to test their study animals for Mycobacterium sp. As has been the case with other free-ranging mammals that come into contact with domestic livestock, there may be public pressure in the future for determining what role, if any, tapirs play in the epidemiology of tuberculosis of domestic animals. Thus it is better to begin researching the accuracy of these new tests and generating preliminary findings sooner rather than later.

3. **Bacillus anthracis.** Although there are no official reports of anthrax in tapirs, Hernández-Camacho (non-official) described a case of the disease in Andean tapirs in Colombia (Downer, pers. comm.). In general, B. anthracis infection in perissodactyls results in sudden death after severe diarrhea with foamy mucous discharge from the mouth and nostrils and eventual rectal prolapse (Ramsay & Zainuddin 1993). This disease and its impact on wild tapir populations must be investigated in endemic regions.

4. **Leptospira spp.** Leptospirosis may be a threat to both captive and wild tapirs. Serological antibody titers against Leptospira in the absence of clinical signs have been reported in wild tapirs (Hernández-Divers et al. 2005; Mangini et al. 2012; Medici et al. 2014). Evidence of clinical disease caused by Leptospira interrogans serovar Pomona was observed in a female lowland tapir in the Brazilian Pantanal, where domestic livestock, such as cattle and horses, live in close proximity to tapirs and other wildlife. Clinical signs included glaucoma, uveitis, low levels of activity and low response to external stimuli (P. R. Mangini, E. P. Medici & J. A. May, personal communication). The relationship between the tapirs and these bacteria and its specific serovars, as well as with the role of tapirs as carriers of the disease must be studied.

It is important to collect samples over time to see if the titers increase. There exists one report of an increase of the titers of several serovars such as Canicola, Gryppotyphosa, Pyrogenes and Wolffi in a wild tapir captured and rehabilitated in a zoo in Mexico (Jonathan Perez, personal communication).

5. **Mandibular swellings.** Tapirs are particularly prone to develop mandibular abscess or “lumpy jaw” both in captivity and in the wild. Although the condition is considered to be similar to that seen in domestic cattle, its pathogenesis in tapirs is unknown.

The microorganisms isolated from the lesions are Corynebacterium pyogenes, ß-hemolytic Streptococcus, Actinomyces, Necrobacillus, Escherichia coli and Mycobacterium. No viruses have been associated

with this disease, but more research is needed to further support initial findings. In fact, mandibular abscess may be a significant problem for wild tapirs as the lesions can affect the bone and lead to osteomyelitis, which frequently ends in death because of systemic involvement. Thus researchers must report cases of mandibular abscess in free-ranging animals and continue to collect samples to identify the pathogens involved.

6. **Clostridium tetani.** Tetanus can be fatal in lowland tapirs, producing muscle stiffness, hemoglobinuria and death in approximately 13 days (Mangini et al. 2002). Some zoo veterinarians perform vaccinations to prevent this disease.

8.1.2. Viral Diseases

Positive serum titers for several viral infections have been reported for wild and captive tapirs. Herpesvirus, encephalomyocarditisvirus and foot-and-mouth disease have been reported as clinical diseases or causes of death for captive tapirs (Ramsay & Zainuddin 1993; Göltenboth et al. 1996; Backues et al. 1999; Janssen 2003; Mangini et al. 2012).

1. **Herpesvirus.** There is one report of mortality in Malayan tapirs as a result of herpesvirus (Janssen et al. 1996). However, the type of herpesvirus was not determined. Little is known about the epidemiology of this disease, even in captive populations. Recently, a new gamma 2 herpesvirus has been partially sequenced in a captive lowland tapir but nothing is known about its potential pathogenicity. As a latent DNA virus, herpesvirus should be common and widespread in populations, but stress and/or immuno-supression (e.g. the effects of fragmented populations, inadequate captive conditions) may reactivate the virus, and lead to clinical (and sometimes lethal) symptoms (de Thoisy, pers. comm.). The Veterinary Committee encourages veterinarians to consult with virologists that specialize in herpesvirus for the appropriate sample collection, analysis and interpretation.

2. **Encephalomyelitis (including West Nile Virus; EEE - Eastern Equine Encephalitis; VEE - Venezuelan Equine Encephalitis; and WEE - Western Equine Encephalitis).** There are no scientific reports that confirm that tapirs are susceptible to encephalitis. However, several zoos vaccinate tapirs for these diseases, and a recent health survey documented serological titers to VEE in a small population of free-ranging Baird's tapirs in Corcovado National Park, Costa Rica, Central America (Hernández-Divers et al. 2005). Additionally, a long-term lowland tapir research project in Morro do Diabo State Park, São Paulo State, Brazil, found positive serum titers for both EEE and WEE (Medici et al. 2014). It is recommended that pre- and post-vaccination titers are performed to determine the efficacy of such vaccines. In addition, any evidence of viral encephalomyelitis should be reported. At Quintana Roo, one tapir was captured and sampled, and was negative to serum titers for EEE, VEE and West Nile Virus (Jonathan Perez unpublished data).

There are anecdotal reports of West Nile Virus clinically affecting rhinos. Therefore, some captive collections of tapirs are currently vaccinated with the equine West Nile Virus vaccine (Ft. Dodge). It is important to obtain pre and post-vaccination titers to West Nile to determine the efficacy of this vaccine. Any tapir that dies as a result of West Nile Virus should be reported.

3. Foot and Mouth Disease. An outbreak of FMD in the Paris Zoo, France, which affected lowland and Malayan tapirs, was described by Urbain et al. (1938). The clinical findings were limited to only interdigital lesions. However, at the Mountain Tapir Population and Habitat Viability Assessment (PHVA) Workshop carried out in Colombia in October 2004, Peruvian field biologist Jessica Amanzo reported two FMD outbreaks in the Northern Peru that produced a high mortality of mountain tapirs. The first outbreak occurred 50 years ago and the second 25 years ago. Although this information has not been confirmed, tapir researchers must continue to monitor for this disease; serologic surveys specific to FMD should be carried out, especially in mountain tapirs.

8.1.3. Parasitic Diseases

The presence of parasites associated, or not, with disease has been frequently described in tapirs. Nevertheless, little data is available on disease manifestation and on the environmental influence on infection and seasonality. In general, parasite presence has not been associated with clinical signs in wild tapirs and reports suggest a certain equilibrium in the parasite-host relationship in natural environments (Mangini et al. 2012). In some cases, the occurrence of clinical manifestations and diseases in tapirs could be related to immune suppressive events. In captivity, parasite infections can represent up to 36% of medical problems (Mangini et al. 2002); and parasite control is recommended, even for asymptomatic captive tapirs with positive results in parasitological exams (Mangini et al. 2012).

Ectoparasites. Ectoparasites are observed in both captive and wild tapirs. In wild tapirs, the identification of ectoparasites such as ticks and flies may offer insight into the interaction between tapirs and livestock, including the risk of mutual transmission of diseases. Identifying parasitic genera that naturally infest tapirs is also of interest. Furthermore, the analysis of ectoparasites can help offer insight into the role that wild tapirs may play as potential reservoirs of some diseases. Amblyomma sp. is the most common ectoparasite reported in tapirs, and can cause dermatological problems (Mangini et al. 1998; Nunes 2001; Mangini et al. 2002). The tick species already recorded on tapirs are the following: Amblyomma brasiliense, A. cajannense, A. calcaratum, A. coelebs, A. dubitatum, A. incisum, A. latepunctatum, A.multipunctum, A. naponense, A. neumanni, A. oblongoguttatum, A. ovale, A. pacae, A. parvum, A. scalpturatum, A. tapirellum, A. pseudoconcolor, A. triste, Haemaphysalis juxtakochi, Dermacentor halli, D. latus, D. (Anocentor) nitens, Ixodes bicornis, I. boliviensis, I. tapirus, I. scapularis, Rhipicephalus (Boophilus) microplus, Ornithodoros rudis and O. tuttlei (Mangini et al.1998; Lira Torres et al. 2001; Labruna & Guglielmone 2010; Medici 2010; Mangini et al. 2012).

Endoparasites. In general, endoparasites have been reported for tapirs numerous times without associated clinical problems, including Parascaris sp., Fasciola hepatica, Capillaria sp., Paranoplocephalasp., Strongyloides sp., Agriostomun sp., Lacandoriasp., Neomurshidia sp., Trichostrongylus sp., Strongylus sp., Brachylumus sp., Eimeria sp., Balantidium sp., and Giardia sp. (Kuehn 1986; Ramsay & Zainuddin 1993; Lira Torres et al. 2001; Pukazhenthi et al. 2008; Santos 2011; Mangini et al. 2012). Naegleria fowleri and Schistosomatidea were reported as pathogenic in captive tapirs (Lozano-Alarconet al. 1997; Yamini & Veen 1988). Some protozoans can be considered as normal enteric flora in tapirs; however they can also be pathogenic in immunos

supressed animals. Giardia duodenalis AI genotype (a zoonotic protozoan frequently reported as causing gastroenteric disease in humans) was detected in a fecal sample of a captive T. terrestris, but was not associated with clinical signs (Santos 2011). Cryptosporidium sp. was described as causing watery diarrhea in two captive T. bairdii in China (Chen et al. 2012). Ascarididae are also frequently found in wild tapir fecal samples in Pantanal (Brazil) (R. C. Fernandes-Santos & E. P. Medici, personal communication). There are some highly pathogenic parasites with cycles in which tapirs may play a central role. One important example is Toxoplasma sp., whose high prevalence has been reported in free-ranging ungulates from French Guiana. Prevalence was significantly linked to terrestria species (de Thoisy et al. 2003), thus tapirs may be infected. We encourage researchers to study endoparasites in wild tapirs in order to further assess the role of tapirs in these cycles and to differentiate those endoparasites that naturally infest tapirs from those acquired by tapirs through interaction with livestock).

Hemoparasites. Tapirs can be parasitized with several species of ticks and other ectoparasites that could be vectors of a wide variety of hemoparasites. Recent reports of hemoparasites in in tapirs include Babesia sp. and Trypanosoma sp. As is the case with endo and ectoparasites, the presence of hemoparasites in tapirs can occur without any clinical manifestation. A wild mountain tapir, Tapirus pinchaque, was recently reported as seropositive for Babesia caballi in Ecuador, without clinical signs (Castellanos 2013). In lowland tapirs Tapirus terrestris in Brazil, a possible new species of the genus Trypanosoma was recently reported, also without clinical manifestation. The species was named Trypanosoma terrestris (Acosta et al. 2013). Although the tapirs did not display any clinical signs in these cases, the real implications of the presence of both hemoparasites in tapir's population health remain unclear and should be carefully studied.

8.1.4. Fungal Diseases

Very few reports are available about fungal diseases affecting tapirs (Manigini et al. 2012). Dermatophytosis caused by Microsporumgypseum, M. canis and Trichophytum tonsurans was observed in tapirs with alopecia episodes (Ramsay& Zainuddin 1993). A fungal coccidioidomycosis (Coccidiodesimmitus) causing respiratory disease was also recently reported (Janssen 2003).

8.1.5. Non-Infectious Diseases

Vesicular Dermatitis. The condition termed "vesicular dermatitis" is the subject of ongoing research in captive tapirs. This condition was first described by Finnegan et al. 1993. Although the syndrome continues to affect captive tapirs, its etiology has still not been identified. To diagnose this syndrome, researchers should perform skin biopsies of affected areas, and collect fluids produced by the lesions then store samples in two ways: frozen and in 10% buffered formalin. A histopathologic examination of the samples is then necessary for proper diagnosis. Further research is necessary to clarify the etiology of "vesicular dermatitis".

Iron Storage Disease. There is some evidence that the iron levels in captive tapirs are significantly higher than in their free-ranging counterparts (Don Paglia, personal communication). This disease has also been reported for black rhinos. Histopathologic evaluation of the tapir's liver is recommended to evaluate whether or not the animal is affected by iron storage disease.

RECOMMENDED LITERATURE

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Picture: Daniel Zupanc

9 Reproduction

9.1. Brief Reproductive Physiology Review

Tapirs have been studied in the wild for over two decades and we have considerable information on tapir ecology, diet, home range sizes and health. Unfortunately little is known about their reproductive biology and physiology, and the data that exist are from tapirs in captivity. Male and female reach puberty between 14 and 48 months of age and in the wild, occasionally the one year-old calves are sighted accompanied by their mothers. Breeding has been observed in females as young as 13 months, and in males as young as 24 months of age. Female tapirs maintain fertility well past 20 years of age and sometimes een past 30 years. A female tapir can have more than 10 offspring in a lifetime (Barongi 1993).

Tapir male have a paired testicle enclosed in a minimally pendulous scrotum located cranioventral to the anus. The testicles are ellipsoidal in shape, and have an average length and width of 10.5 and 4.8 cm in the Baird's tapir (Pukazhenthi et al. 2011), and 9.7 and 5.0 cm in the Malayan tapir (Lilia et al. 2010). Testis size and volume varies among males depending on age. Adult male tapirs have pronounced cauda epididymides, which can be indicative of successful sperm production.

The penis is typically retracted into the prepuce, but the glans is always visible and directed caudally. Near the glans penis, there are three flap-like projections (erectile tissue), two located laterally and one dorsally. The accessory glands are comprised of a pair of vesicular glands located near the neck of the bladder; a paired prostate located caudal to the vesicular glands; and a bulbourethral gland located about 3.0 cm caudal to the prostate gland that opens directly into the urethra (Pukazhenthi et al. 2011).

To urinate they move the extremity of the penis backwards, and propel the urine a considerable distance away from themselves. Like in the domestic horse, the tapir's urethra finishes with a small prominence in the lower side of the gland. From the penis morphologyin erection, it may be deduced that the ejaculation occurs inside the uterus, like in equines.

The reproductive anatomy of the female tapir appears to be similar to that of the domestic horse and rhinoceros. It has been described in detail only in the Malayan tapir (Lilia et al. 2010). The reproductive tract is comprised of the external genitalia (vulva, clitoris), vagina, cervix, uterus, oviducts and a pair of ovaries. The vulva has two labia that are sparsely covered with hair and both the dorsal and ventral commissures are rounded. The vulvar opening is a vertical slit measuring about 4.8 cm in length. The clitoris is located on the ventral floor of the vaginal vestibule about 1.0 cm cranial to the vulva. The vagina and vaginal vestibule are thin walled with smooth mucosal layers. The vaginal mucous produces a lipid secretion that seals the vulvar lips, isolating the vaginal environment from external medium and protecting it when the animal stays in the water.

The cervix is firm and muscular and measures about 3.0 cm in length and 5.3 cm wide in the adult, is located dorso-cranial to the urinary bladder and anterior to the brim of the pelvis.The uterine body is relatively short (16.8 cm, sub-adult; 7.3 cm, adult) compared to the uterine horns. The average length of the uterine horn is 31.2 cm and the average width is 2.8 cm. Tapirs have two uterine horns. The shape of the ovaries is oval to elongate; they are located on the ventral surface of the ilium near the coxal tuber. In adult females the average length is 2.8 cm and width is 1.3 cm. Tapirs have an epitheliochorial and diffuse placentation (Pukazhenthi et al. 2011). Females have a pair of mammary glands on the inguinal area.

Tapir estrous is very difficult to determine. In general, female tapirs are polyestrical over the course of a year. Estrous lasts 1-4 days and is repeated every 28-32 days. Fertile estrus is possible 9-27 days postpartum. However, these figures may vary by species and the environment of individual tapirs and should be used as general guidelines.

9.2. Hormones during estral cycle and gestation

Hormone screening is used to monitor the estrous cycle and hormonal status in both captive and free-living animals. Because of the stress produced by immobilization, blood samples are not reliable for studying hormones in captured individuals. In untrained, captive females, urine samples are the best choice for diagnosing and monitoring pregnancy. The collection of urine is minimally invasive and thus allows for more accurate measurements of hormonal concentrations given that the animals are not stressed out by the capture process. Fecal samples are the best choice for field studies. Fecal samples to be used for hormone screening must be collected immediately after defecation. The samples can be stored in a container with ethanol 90% and the precise time of collection must be recorded. The sample can be dried in an oven, sunlight, or a lyophilizeror extracted in the field as mentioned earlier. Given the logistic difficulty of collecting fresh feces samples in some field conditions, it is important for veterinarians to continue improving other non-invasive approaches, like fecal steroid in order to determine estrous cycle and pregnancy in tapirs for wild individuals (Pukazhenthi et al. 2013).

The most widely employed analysis for the detection of hormone metabolites is the radio-immunoassay. In captive, trained tapirs, samples should be collected at least weekly in order to have the baseline data on the fluctuations of progesterone serum levels needed to confirm pregnancy. Ultrasounds can also be used to confirm pregnancy.

Research on the serum progesterone concentrations of captive Baird's tapirs conducted by Dr. Janine Brown (1994) in the United States indicates that the duration of the estrous cycle is about 25-38 days, with a luteal phase length of 15-20 days. The interluteal period is relatively long, comprising approximately 40% of the estrous cycle. Females resume cycling 16.2±2 days after parturition and can become pregnant during the first postpartum estrous.

Hormonal screening carried out with captive lowland tapirs by Fundación Temaikén, Argentina, showed that the serum estrogen concentrations vary between 17.2-35.1ng/ml, and serum progesterone concentrations vary between 0.78-1.64ng/ml. The male serum testosterone concentrations vary between 0.12-1.73ng/ml; a concentration of 0.2 ng/ml was registered during the copulation period in a male of lowland tapir.

Copulation can take place on land or in the water. The gestation period for tapirs is quite long and varie by species. For Lowland tapirs it is approximately 395 to 399 days, while it is shorter for Malayan tapirs and Baird's tapirs. Even in the latter stages the gestation is not detectable using a physical or visual test. As indicated above, gestation must be confirmed by ultrasound or through an analysis of hormonal concentrations in serum, urine or feces. Little is known about tapir vaginal cytology, but recent data suggest that it might be possible to use cytological research to differentiate the stages of the estrous cycle stages and to diagnose gestation.

Progesterone concentrations higher than 2.5ng/ml are likely to indicate pregnancy, but veterinarians should conduct 3 tests during a 15 period to make a definitive diagnosis. If the progesterone values increase over the course of the three tests, the tapir is pregnant and the veterinary staff can shift their focus to monitoring fetal development.

In pregnant lowland tapirs, progesterone serum concentrations fluctuate throughout the gestation period, registering minimum values of 2.67ng/

ml during the initial stage and maximum values of 22.6ng/ml during the final stage. In contrast, estrogen serum concentrations show uniform behavior throughout gestation and typically measure 20-30pg/ml. In lowland tapirs research shows that both hormones reach a maximum level 7-10 days before parturition, but then decrease drastically in the last few hours before parturition (Quse et al. 2004). A similar process has been described in Baird's tapirs, with estrogens values considerably higher, of 85-131 pg/ml (Brown et al. 1994).

In lowland tapirs cortisol does not seem to play an important role in initiating parturition, since its serum concentrations do not show significant changes toward the end of gestation. The values registered during pregnancy varied from 2.52ng/ml in the first period of gestation to 3.19ng/ml 48 hours before parturition. This pattern has also been observed in Baird's tapirs. In the early stages of gestation, their cortisol concentrations varied between 6.9-10.2ng/ml; in the later stage, the values were between 9.5-10.8ng/ml (Quse et al. 2004).

Given the minimal data on the gestation and fetal development of wild tapirs, conducting ultrasound exams in the field would produce valuable, novel information. For tapirs in captivity, ultrasound exams can be conducted with the female in a standing position or in lateral recumbency. In the wild, anesthesia is necessary. Transabdominal ultrasonography is the method of choice for the diagnosis of pregnancy with a transducer of about 3.5 MHz. The abdominal region is spread with gel or perfectly moistened with alcohol in order to minimize interference from the hair coat (Fernandez Jurado, personal communication; Hoyer et al., 2007; Van Engeldorp Gastelaars, 2010). Before the third month of gestation, it is necessary use a 3.5 to 5 MHz transvaginal transducer in order to see the gestational sac (Fernandez Jurado, personal communication). A Doppler or real-time (B-mode) ultrasound equipment can also be use for checking fetal movements and heartbeat. Attaching the probe to extensions made of PVC pipe permits easier introduction of the transducer into the rectum and deeper access (Pukazhenthi et al. 2013).

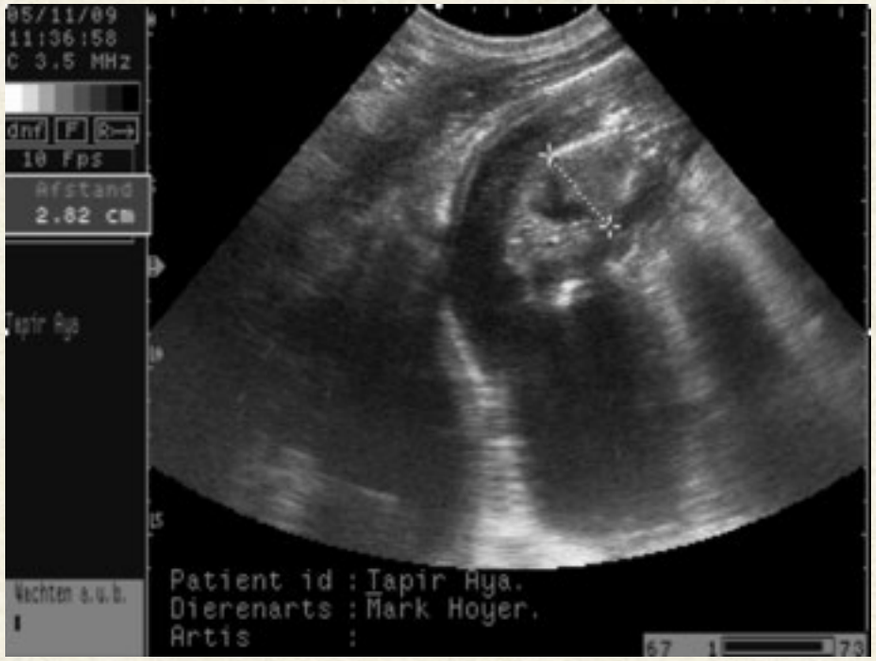
The following there are several ultrasound pictures of various lenghts of pregnancy of a Malayan tapir (Tapirus indicus), from Artis Zoo (Holland). Pictures were obtained transabdominally using a 3,5 MHz convex probe (Esaote PIEmedical Aquila®) or 2,5 – 6,6 MHz convex probe (Esaote MyLabOne Vet, Esaote®, Esaote Benelux BV, Maastricht, the Netherlands). The ultrasound was conducted with a trained animal in lateral recumbancy (Hoyer 2014).

For full details, please refer to M.J Hoyer and H.D.M Post – van Engeldorp Gastelaars: Ultrasonic characterization of fetal development in a captive Malayan tapir (Tapirus indicus). Accepted for publication in Zoo Biology 2014.

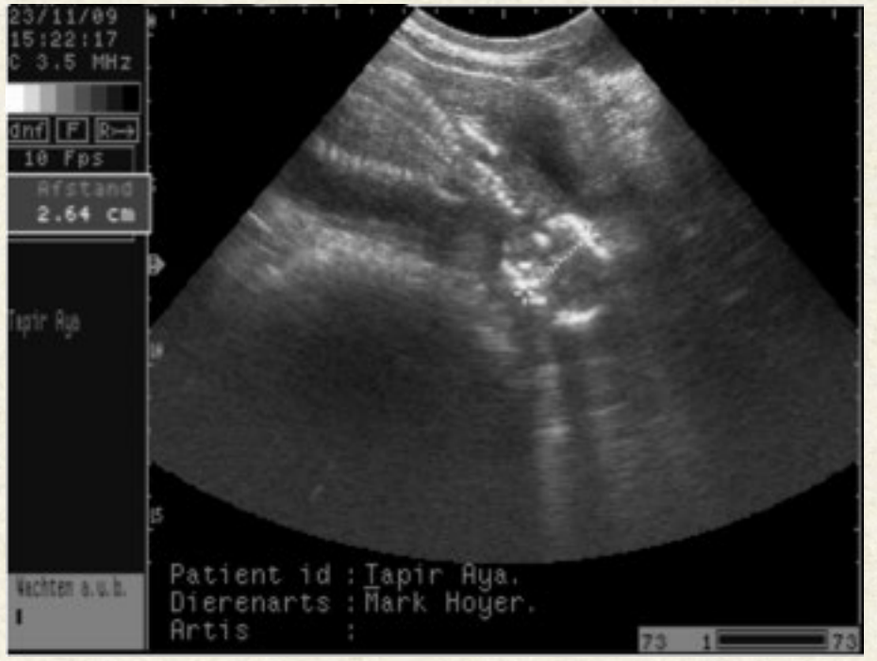
The recommended measurements to monitor fetal development are the biparietal and thoracic diameters and total length of the fetus. Studies with 3-month old lowland tapir fetus showed a biparietal diameter of 2.35 cm and body length of 15 cm. At 6 months, the biparietal diameter was 3.02 cm, the thoracic dorsum ventral length was 6.5 cm and the total length was 20 cm. At the end of the pregnancy the fetus measured 75 cm long, with a biparietal diameter of 11 cm and a thoracic diameter of 40 cm (Fernandez Jurado, personal communication).

In the earliest stages, it is difficult to diagnose pregnancy using an ultrasound exam given the thickness of the abdominal wall in adult tapirs and the minimal clinical signs of pregnancy.

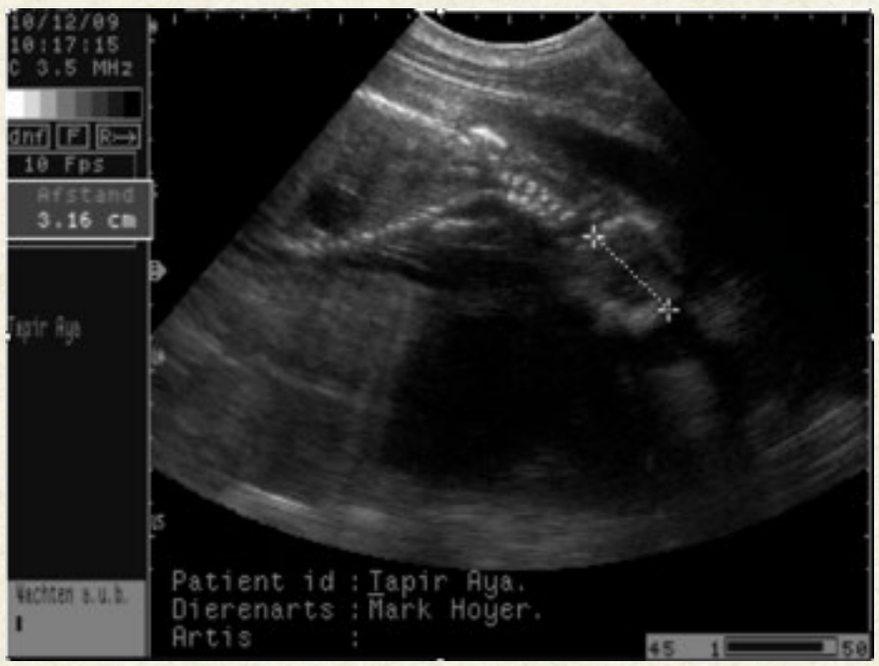
Gallery 10 - Reproduction: Ultrasound exam



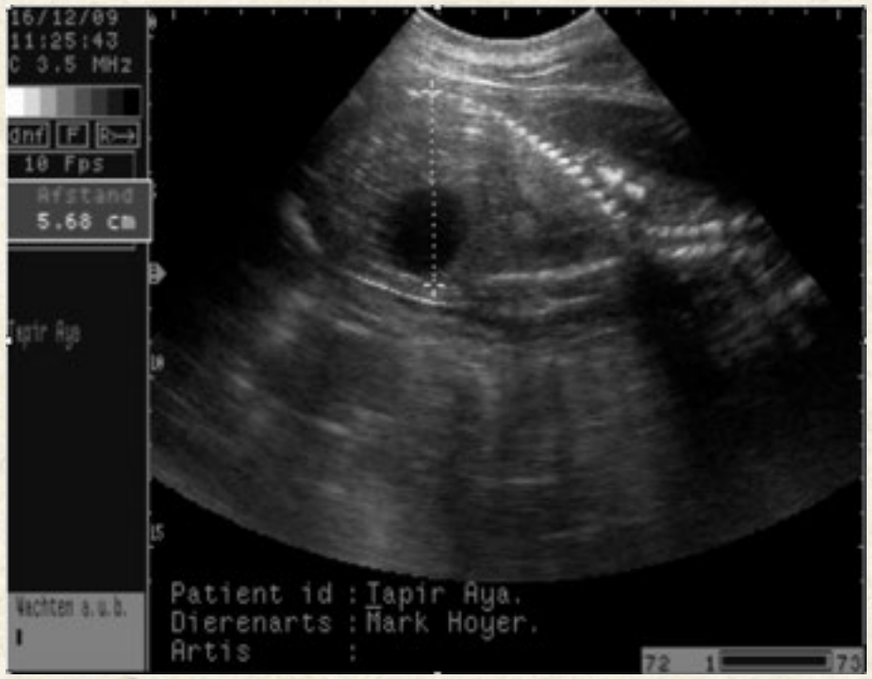
Tapirus indicus 3 months old fetus. Cross thorax diameter 2.82cm. Image and photo: Mark Hoyer.



Tapirus indicus 3.5 months old fetus. Head, neck, thorax and front legs clearly visible. Bioparietal diameter 2.64cm. Image and photo: Mark Hoyer.



Tapirus indicus 4.5 months old fetus. Stomach and heart clearly visible. Image and photo: Mark Hoyer.



Tapirus indicus 4.5 months old fetus. Rib cage. Image and photo: Mark Hoyer.

9.3. Recommended Research Topics

There is a dearth of data on tapir reproductive physiology and the TSG Veterinary Committee views improving our knowledge in this area of tapir health as a top priority. Given the comparative ease of researching the tapir reproduction of captive versus wild tapirs, the most effective approach in the short term is to research captive animals with methodologies designed to yield data that will contribute to conservation efforts. The following is a list of research topics within tapir repductive physiology that the TSG Veterinary Committee believes to be of utmost importance:

1. Monitoring of reproductive hormones through non-invasive methods;

2. Electro-ejaculation and sperm handling and storage, accompanied by studies of spermatozoid viability;

3. Artificial insemination protocols;

4. Collection, preservation and viability analysis of oocytes;

5. Monitoring fetal viability using ultrasound studies (most feasible with train, captive tapirs);

6. Nutritional requirements for the pregnant female during different periods of pregnancy;

7. Analyses of the nutritional composition of milk (including colostrums), in the four tapir species.

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Picture: Diego Lizcano

10 Necropsy

Necropsy

Field necropsies yield valuable information on wild tapir health. They are rare opportunities that should never be wasted. It is more common to find carcasses in advanced stages of decomposition rather than fresh corpses, but given the possibility fo collecting important data, even in these cases veterinarians should conduct a limited necropsy evaluation. It is difficult to refrigerate or freeze adult tapir carcasses, so field necropsies must be carried out efficiently.

Proper protective gear is necessary to conduct any necropsy, including disposable latex gloves, a mask, and protective eyewear, clothes and boots. Standard surgical masks do not provide adequate protection against potentially zoonotic agents such as Mycobacterium tuberculosis, thus N95 masks are recommended for tapir necropsies.

The necropsy is essentially an exercise in observation and description, and should involve little interpretation unless the veterinarian performing the necropsy is an experienced pathologist. Throughout the necropsy, the veterinarian should record an accurate and detailed description of the appearance and texture of the tissues. It is necessary to record particularly detailed decriptions upon viewing potential anomalies. It can be extremely useful to photograph the necropsy, as photos allow the later re-evaluation of data and can permit experienced pathologists to offer second opinions.

The objective of a necropsy is to document the state of the tapir's health prior to death, including the processes that led to the death of the animal and all the others that occurred concurrently. To achieve this, all tissues and organs should be carefully observed and sampled for bacteriology and histopathology. The collection of gastric content, parasites, genetic samples, etc. is useful to provide a basis of comparison with other

animals with unknown cause of death and to provide basic data on the biology of the species.

Necropsy notes should avoid subjective or colloquial terms (a lot, much, few, huge, etc.) and should use objective descriptions and precise measurements whenever feasible. The use of necropsy protocols to ensure that data collection is adequate. APPENDIX 2 provides a spreadsheet and a simple checklist for field necropsies. We encourage researchers to use this spreadsheet to help standardize the information collected by different tapir research projects, which will facilitate comparisons on the causes of death of tapirs in different locations.

All instruments and equipment used in the necropsy must be disinfected, and wastes stored and disposed of following national biosecurity regulations (see Chapter 6).

The necropsy classically is divided in three phases:

1. External exam (skin, mucosa, natural orifices, apparent health).

2. Structural organization of viscera (compression, volvulus, dystopias, cavitary liquids).

3. Individual evaluation of the organs.

In case that there is the opportunity to perform a necropsy, the following basic recommendations may prove helpful:

– Tapir is placed in right lateral decubitus (or left lateral decubitus if the person conducting the necropsy is left-handed) to facilitate the removal of different organs.

– In order to open the carcass, it will be necessary to make an incision along the midline from the chin to the anus (in females through the middle of the mammary glands).

– Divulse the skin up to the back line of the body, folding it so to expose the ribcage and abdominal musculature. Cut from the forelimb below the scapula and in the hindlim muscle mass inside of the leg opening the hip joint and cutting the round ligament in order to both members can be dumped on each side of the body. Bind the large blood vessels in order to avoid hemorragy in case cut it.

– Cut the ribs and the diaphragm and then remove all costal and abdominal walls in a one piece. This is help as a clean area in order to put the organs removed from the cavity (important in the field).

All organs should be analyzedin relation to their external (size, form, location, surface, color, symmetry) and internal characteristics (structure, consistence, content, thickness, parasites, cutting surface, internal color, symmetry, nodules). Each lesion and abnormality should be meticulously evaluated; if unclear whether the tissue is abnormal, describe it as throroughly as possible.

Tissues and organs should be photographed extensively, regardless whether they appear normal or abnormal. The series of photographs should illustrate the organ's size, position, texture, content, etc. Photographs should include a ruler or some size reference (e.g. scalpel), should be clearly focused and, as much as possible, photographs should be at an angle perpendicular to the tissue being photographed. Natural

light is preferable over flash photography, but if proper light cannot be obtained do not hesitate to use flash.

It is useful to begin by thoracic cavity to avoid contamination could potentially result if we work first with the abdominal organs and intestines are ruptured. The following order necropsy procedures are suggested:

– Before removing all the organs from the thoracic cavity, observe the heart and lungs in situ. Evaluatetheir size, shape and position; verify if there are points of adhesion or fluids insidethe pericardial sac. In case of presence of liquid, remove it with a sterile syringe (refrigerate and submit to microbialculture) and evaluate and describe the liquid'scolor, transparency, and volume. The same applies to the visceral and parietal pleura. It should be considered, however, that in T. indicus the parietal and visceral pleura are normally thick and prominent, and there is a fibrous connective tissue between the lung and chest wall that might be mistaken for pathological adhesions.

– Observe mediastinal lymph nodes, thryoid, parathyroid and, if present, the thymus, evaluating their size, color and texture; cut these organs in order to evaluate their internal macroscopic characteristics.

– The thoracic organs are removed together with the tongue, larynx, trachea and cranial esophagus, starting by the tongue.

– The trachea should be opened with scissors in its entirety to the bifurcation of the main bronchi to evaluate for the presence of parasites, foam (edema and pulmonary emphysema), or exudates (pneumonia).

– The lungs should be examined for the presence of nodules, abscesses, emphysema, edema, congestion. Cut the parenchyma and note if there is liquid (evaluate the color and characteristics). Open bronchi and observe presence of liquids or parasites.

– Evaluate abdominal organs in situ.

– Observe mesenteric lymph nodes (size, color); cut the nodes in order to evaluate their internal macroscopic characteristics.

– Examine the bladder (a urine sample may be collected aseptically for culture if necessary), and then evaluate the bladder mucosa.

– Remove the spleen and pancreas and evaluate their size, surface, color and consistency. Cut the parenchyma in different parts to examine.

– Remove the stomach using double ligatures in the cardia and pylorum (perform 2 ligatures at each anatomic site then cut between the two ligatures in order to avoid spreading the organ's contents). Open the esophagus (with a scissor) to observe the characteristic of mucosa, lesions or presence of foreign bodies. The stomach is best opened by its lesser curvature, to prevent its contents from spilling; following the inspection and photographing of the contents, they may be discarded to allow for the evaluation of themucosa.

– To remove the liver it is best to bind its large blood vessels, preventing excessive blood outlet. Note the size, color, consistency, presence of lighter or darker areas than the normal

color pattern and characteristics of the liver edges. Cut the organ in successive sectionsto evaluate the liver parenchyma.

– The best way to remove the intestines is by releasing the mesentery with the intestines inside the body: hold the scalpel or knife at an angle of approximately 45 degrees to the mesentery, and gently pull on the intestines along the scalpel to cut free it from the mesentery. To remove the intestine it is best totie it with a string on the proximal duodenum and distal rectum (double ligatures are advised). Evaluate the intestinal content, mucosa color, presence of parasites, etc.

– Remove the kidneys and adrenals. Split the kidneys longtudinally through the greater curvature to the hilum to observe the characteristics of the cortex, medulla and and pelvis (in adult tapirs the cortex should represent approximately 80% of the renal mass). The renal capsule should be carefully removed to determine if there is adhesion and to observe the outer kidney surface.

In case there is suspicion of neurological disease, the brain should be the first to be examined, in order to minimize autolysis.

Following necropsy, the carcass should be submitted to appropriate disposal services or buried in order to avoid contamination.

The necropsy provides the opportunity of collecting a series of samples for posterior laboratorial exams, as summarized in TABLE 7.

Table 10 - *Collection, Handling and Storage of Samples from Necropsies.*

Analysis	Objective	Sample	Collection and Handling	Storage
Histopathology	Complements the necropsy, identifying pathological processes and the cause of death.	All organs should be collected, altered or not.	Fragments should be no larger than 1 cm^3, always including a fraction of normal tissue. Use a clean flask with formol 10% (formalin 4%) in a volume 8-10 times greater than the samples.	Keep the flasks well closed and safe from light, at ambient temperature. These samples will be valid for years.
Microbiology	Identify bacterial or viral agents involved in infectious processes.	Collect only samples from tissue/liquids suspected of infection, soon after the death.	Puncture (1-3mL) for liquids or swab for tissues and abscesses. The assepsy of the procedure is essential.	Keep in sterile flask (or inside the syringe used for puncture) or in nutritive transport media (*e.g.* Stuart) under refrigeration. Send to laboratory within a few hours.
Toxicology	Identify if the animal was exposed to a toxin (environmental contamination, poisoning).	All the viscera (or at least brain, lungs, liver, kidneys and bone marrow), stomach content, hair, fat and cardiac blood should be sampled.	Large fragments (~100g) of the tissues and stomach content, heart blood puncture (~50mL) and hair (store in envelope).	Keep the flask under refrigeration or freezing. Send to laboratory within a few days.
Ectoparasites	Identify the ectoparasites.	Any parasites found on the skin, 5-20 individuals of each apparent species.	Transfer the parasites to a perfored flask (for longer periods, add leaves or wet cotton) or to ethanol 70%.	Keep the flask at ambient temperature. Send to laboratory within a few days (perfored flask) or weeks (ethanol).
Endoparasites	Identify the endoparasites.	Any parasites found on the viscera, 5-20 individuals of each apparent species.	Wash the parasites in water and transfer them to ethanol 70% (cylindrical worms) or AFA (flat worms).	Keep the flask at ambient temperature. Send to laboratory within a few weeks.
Stomach Content	Identify the feeding habits of wild animals.	Stomach content.	Transfer all the stomach content to a bucket, homogenize and collect several small samples to a total of 500mL or 1L.	Keep at ambient temperature or under refrigeration. Use filtration, decantation or greenhouse to dry the sample.
Testicles and Ovaries	Store gametes for assisted reproduction techniques or germplasm banks.	Testicles or ovaries, only from very fresh corpses (<6-18 h).	Collect the intact gonads, donot dissecate them from their serous membranes.	Refrigerate or freeze with maximum urgency, depending on the technique to be applied. Send to laboratory with maximum urgency.
Genetic Analysis	Check IUCN/SSC Tapir Specialist Group (TSG) Manual of Sampling Techniques for Genetic Analysis			
Taxidermization	Consult local museums and taxidermists or the appropriate literature to obtain specific recommendations on the preparation of taxidermized animals or parts. Consult also the local laws of transport and use of tapir parts and the current CITES regulations.			
Other Analysis	The other samples described on the chapter “Collection, handling and storage of biological samples” such as corporal measurements, hair, feces, urine and other, may also be collected following the same recommendations.			

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Picture: Daniel Zupanc

11

Interventions in Individual and Population Health

Chapter 11

Interventions in Individual and Population Health

Intervening in wild animal population health is very controversial. The decision to conduct any therapeutic or prophylactic intervention, must be made after considering the possible effects on the balance of the local ecosystem, the conservation of the species and on-going evolutionary processes. There is no single rule for whether a veterinarian should or should not intervene in the health of a wild animal. However, whenever the choice is taken to intervene, the veterinarian must make sure that this action will imply no risk to the survival of the rest of the population or to the stability of the ecosystem (e.g. live vaccines, resistant bacteria selection, etc.).

There is a general consensus that veterinarians should treat lesions inflicted on study animals during their capture, immobilization, or processing. Examples include trap related injuries, injuries caused by hunting dogs, and chronic lesions from radio-collars. The treatment of non-capture-related lesions is much more controversial. One of the pillars of the conservation philosophy is to make sure that the evolutionary process continues in its natural balance, and some argue that the treatment of these lesions interferes with the natural processes of mortality and evolution and is therefore not recommended. For instance, wounded animals may be an important part of large predators' diet and treating wounded animals can harm these predators. In contrast to this, others argue that most of the injuries observed in wild tapirs are probably indirect consequences of human interference in the animal's habitat, and thus treating these lesions actually minimizes the net anthropogenic interference in local ecosystems. Another argument for treating all lesions is that in reduced populations and endangered species, the death of a single individual might have important

consequences on the population. Thus, treating injured animals may contribute to the survival of the species.

The question of whether or not veterinarians should intervene in these contexts has no clear answer and will vary depending on the particular case. Therefore in this chapter, rather than attempting to provide any kind of definitive answer, we instead offer a list of important variables and clinical signs along with their potential consequences that a veterinarian should evaluate before deciding whether or not to treat captured wild tapirs with wounds not related to the capture process.

The most common clinical signs observed in wild tapirs include:

-High and medium infestation by ectoparasites;

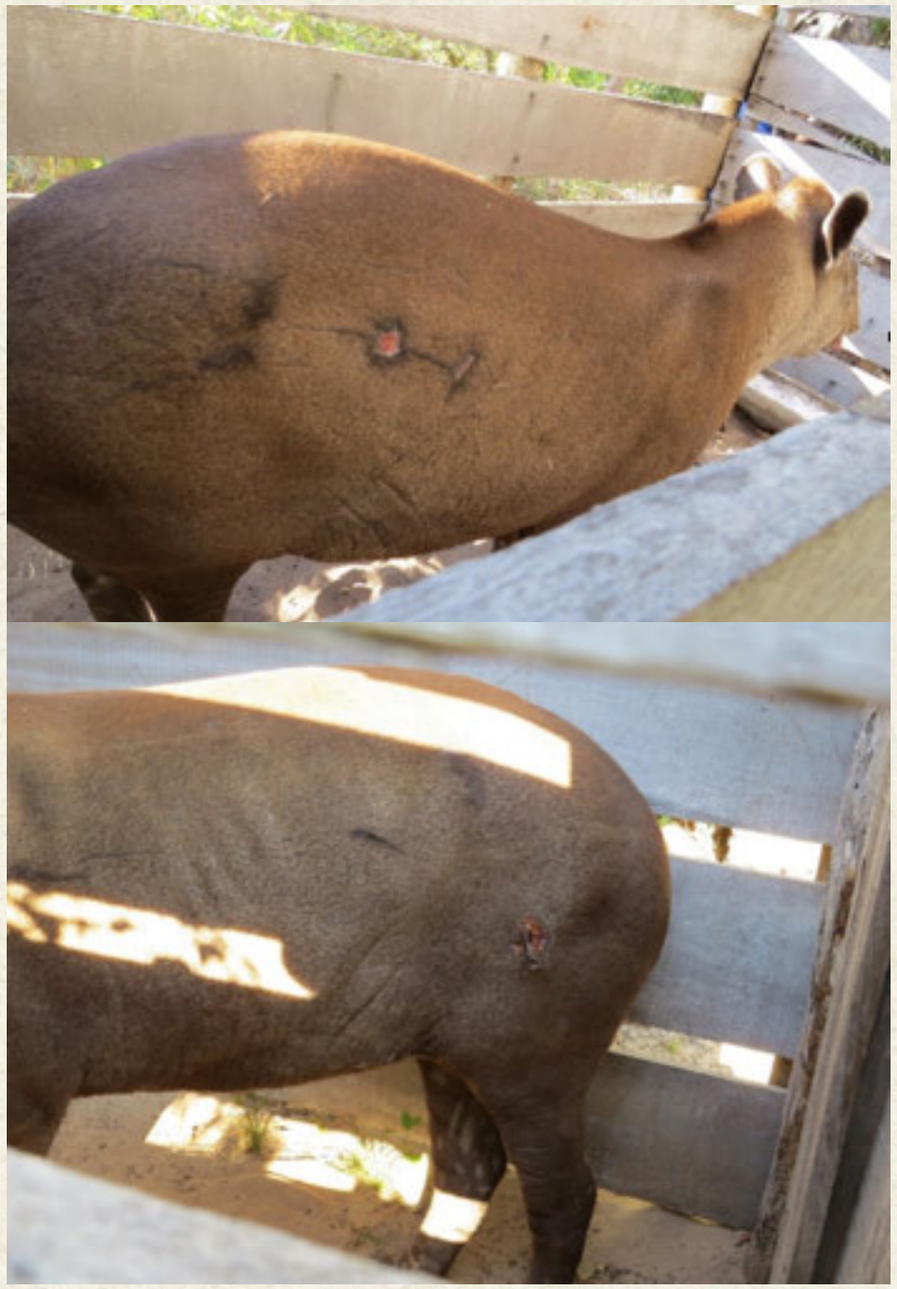
-Skin lesions;

-Myiasis;

-Low body score (thinness, cachexia);

-Presence of secretions in natural body cavities (particularly ocular, unilateral or bilateral).

Gallery 11 - Examples of skin lesions



Examples of skin lesions not uncommonly observed in wild lowland tapirs. Photos: Renata Carolina Fernandes-Santos.

Gallery 12 - Example of low body score (caquexia)

Wild lowland tapir in Brazilian Pantanal showing very low body score (cachexia). Photos: Lowland Tapir Conservation Initiative (camera trap records).

Other less common clinical signs already observed in wild lowland tapirs include: lameness, alopecia, phlegmon, dental fractures, eye conditions (senile halo, lens opacity, corneal lesions, and periophthalmic glandular inflammation), and abnormal vaginal discharge.

In most cases, physical abnormalities in wild tapirs can be explained by age (e.g. tooth wear, ocular senile halo) or social behavior (e.g. scars, wounds) rather than disease. Adult tapirs present significantly more scars and/or wounds compared to sub-adults and juveniles. This likely reflects a buildup of scars over the course of an individual's life, but could also suggest an increase in agonistic intra-specific (territoriality) and/or inter-specific (predation) interactions in adults, or that adults survive predation better than younger individuals (Medici et al. 2014).

Additional important factors to be considered before clinical interventions include:

- Overall condition of the individual (overall body condition, locomotion ability, behavior).

- Presence of nonspecific clinical signs.

- Presence of specific clinical signs, or suggestive signs of certain diseases or respiratory and gastrointestinal distress.

- Simultaneous occurrence of more than one specific/nonspecific clinical sign (e.g. high infestation by ectoparasites, cachexia and apathy in the same individual).

- Severity/chronicity of clinical signs.

- Environmental conditions that can influence the overall condition of the individual (such as seasonality and availability of food resources).

- If there is (or was) similar cases (individuals with the same clinical signs) in the population of the same area.

- Results of previous health assessments (individual and population).

It is important that the field veterinarian carries a basic kit of medicines and basic equipment for clinical procedures in case it is necessary to perform some medical intervention. This includes:

- Antiseptic solutions for cleaning and desinfection: alcohol 70%, iodinated alcohol, povidona, clorhexidine 2% or 5%, clorhexidine saline solution 0.2% (0.9 % NaCl).

- Broad spectrum and long action antibiotics (for a single dose). Keep in mind that applying large doses of antibiotics in the wild is quite controversial, since there exist very little data on the potential impacts and may lead to strains of bacteria that are resistant to common antibiotics.

- Steroidal and non steroidal anti inflammatories.

- Fluid therapy solutions and equipment (e.g. 0.9%NaCl).

- Eye drops or ointment for cleaning and treatment of ocular injuries.

- Antimicrobial or antimicotical ointments or creams to treat skin lesions caused by bacteria or fungi.

- Topical antiparasitic (myasis) and systemic use.

- Multivitamins (injectables).

- Kit of basic surgical instruments: scissors, tweezers (dissection, tooth mouse, etc.), bistoury sheets and mango, gloves, suture thread and needles, swabs, chinstrap.

- Gauze and adhesive tape.

If the veterinarian decides to intervene, the treatment must be accompanied by the collection of biological samples appropriate to conduct subsequent diagnostic exams. These data will help guide decisions about future interventions, and contribute to ur understanding of which pathogens/diseases play a major role in wild tapir population health.

As was the case with necropsies, it is important for field veterinarians to use approproate personal protective equipment (PPE) and give attention to biosafety norms in collecting samples and dealing with bodily fluids and wastes and other potentially contagious materials.

It is important to consider that clinical manifestations in the wild can have very different profiles and sources than that the same clinical signs in captivity, so treatment in wild specimens may differ from those used in captive tapirs. Before deciding on a specific treatment, the veterinarian should consider some of the unique aspects of field veterinary work, including the inability to reevaluate the patient's condition, which implies that the medications should be effective in single doses. In free-living animals, therapeutic protocols are typically based on limited clinical evidence and, in general we recommend the use of supportive therapies rather than complex therapeutic procedures. This is especially true given the general lack of information about wild tapir health. Indeed, it is still unknown which diseases, if any, play a major role in the dynamics of wild tapir populations. Thus it is difficult to justify complex medical procedures designed to cure particular diseases or conditions. Given this context, whether or not a field veterinarian chooses to treat a wild specimen, it is extremely important that field veterinarians compile records of sick animals and report the occurrence of clinical signs and mortality in the wild.

Recent records (2014) of cachectic Baird's tapirs were observed in photos taken by tourism guides in Corcovado National Park in Costa Rica. In cases like these, it is imperative to identify experienced professionals to carefully assess the need for emergency response plans, including the capture, chemical restraint, clinical assessment and collection of biological samples for laboratory analyses.

Of particular note to this chapter are records from 2005-2007 that include the death of 19 adult lowland tapirs, all apparently healthy (with good body mass), in the region of Madre de Dios, in Peru and Northern Bolivia. The cause of death could not be determined due to the difficulty of obtaining fresh biological samples. After the initial reports, the Frankfurt Zoological Society (FZS) and SENASA (Animal Health Service of Peru) signed an agreement in which FZS technicians working in the region of Madre de Dios trained local rangers to collect biological samples from dead animals, which were then sent to the SENASA laboratories in the town of Puerto Maldonado for analyses. These professionals attempted to develop a protocol to reduce the time

between the collection of samples in the field and the processing of the sample in the laboratories in town (Renata Leite Pitman, personal communication, 2014). Unfortunately, the cause of death continued undefined due to logistical issues. The researchers involved in the case suspected West Nile Virus due to ancillary evidence recorded at the same time, including bird deaths, and previous data. A secondary hypothesis was intoxication given that the tapirs were in good physical condition and had recently eaten, indicating that it was an acute death (Renata Leite Pitman, personal communication, 2014).

Gallery 13 - Deaths records

Deaths records of wild lowland tapirs in the region of Madre de Dios, in Peru and Northern Bolivia, between 2005 and 2007. Photos: Renata Leite Pitman

The removal of individuals of high genetic value may be considered during high epidemic risk situations. These individuals may be transferred to captivity or low risk areas, following the recommendations detailed in the IUCN/SSC Tapir Specialist Group's (TSG) Experimental Protocols for Tapir Re-Introduction and Translocation (www.tapirs.org).

In general reintroduction and translocation programs are important wildlife conservation strategies, as well as aggressive intervention in the status of populations and the ecosystem, especially in highly fragmented and threatened populations. Reintroduction and translocation programs both require special attention to the health of selected individuals and the local ecosystem. It is possible that animals included in these management strategies will require therapeutic interventions before and after release or translocation, thus many of the topics in this chapter will be of relevance.

Vaccination protocols, if necessary, should be applied carefully, using only inactivated vaccines, or vaccines that have been previously validated for tapirs or other ungulates. Diseases where vaccines might be justifiably used include tetanus, Infectious Bovine Rhinotracheitis, and Equine Encephalomyelitis.

NOTE: The Tapir Specialist Group -through their Veterinary Committee- asks all those working with tapirs in the wild or in areas where tapirs share habitat with other wild or domestic species to inform the TSG Veterinary Committee immediately of any zoonotic disease outbreaks that jeopardize the lives of tapirs. It is important to consult with our specialists to determine the best course of action. You can get in contact with: Renata Carolina Fernandes Santos: renatacfsantos@gmail.com.bror Viviana Quse: vivianaquse@gmail.com; members of the TSG Veterinary Committee.

Picture: Taipei Zoo

12 Husbandry Guidelines

Husbandry Guidelines

Tapirs are commonly held in zoos and bioparks around the world. In general they are fairly easy to keep and do well in captivity as long as their keepers consider their physiological and biological needs and social behavior.

While certain tapirs are extremely calm, others can be very aggressive towards other animals or keepers. Tapirs are powerful animals and sometime exhibit an unpredictable behavior. Indeed, there are several reports of tapir attacks on keepers, veterinarians, or field researchers that resulted in serious wounds including the loss of fingers.

Thus keepers and zoo authorities must respect tapirs, and design management plans and precautionary measures to minimize risks to tapirs and personnel working directly with them. Adhering to well established husbandry guidelines based specifically on the inter- and intra-specific social behavior of tapirs, the age and sex of tapirs and the subsequent affects on behavior, tapir nutritional needs, spatial requirements, breeding behaviors and requirements, and tapir health will help to minimize the risk of accidents or attacks.

In this chapter, we present some basic management guidelines for those people working directly with tapirs in captivity. These guidelines are intended both to reduce the inherent risks of working with tapirs and improve the quality of life of captive animals. These guidelines are based on the Husbandry Guidelines for Keeping Tapirs in Captivity (Barongi 2000; Shoemaker et al. 2003), the AZA Tapir Care Manual (2013) and the experience of the people who work with tapirs in captivity in different zoos around the world.

We define ‘tapir enclosure’ as both the Exhibit and Holding Areas.

– Each stall of the indoor facilities should have a minimum dimension between 10 m2 to 17m2 (107 ft2 to 180 ft2), interconnected with 1.20m to 1.52m (4ft to 5ft), wide sliding gates that can be operated without physical contact between keepers and tapirs. It is important to have one stall for each tapir so that they can be separated due to behavioral problems, diseases, or for parturition.

– The walls of the indoor facility should be a minimum of 2m (6ft) high and ideally constructed of wood or concrete. Other options are vertical steel bars with less 20 cm (8 in.) spacing in order to prevent tapirs from climbing.

– Floor surfaces should not be rough in order to avoid abrasions to tapirs' soft footpads, which can result in chronic lameness and other foot problems. Zoos in cool or cold climates should have heating systems or provide adequate bedding to insulate against cold floors.

– Some institutions use bedding materials such as grass hay or pine shavings to cover slick surfaces in indoor facilities. Thin layers of bedding may exacerbate the hazards posed by slick surfaces, thus a sufficiently thick layer mus tbe applued. If hay is used as bedding, coarse hay should be avoided, as tapirs are prone to lumpy jaw syndrome if it is ingested. There are several commercially-available synthetic flooring systems that are proving durable in large animal enclosures, and can withstand daily cleaning with disinfectant agents. The floor of indoor facilities should be slightly sloped toward large covered drains.

– The indoor temperatures should be maintained between 16ºC to 29ºC (65 to 85 degrees Fahrenheit); as mentioned above, cold weather zoos should have heated systems or heated floors and the temperature should be monitored throughout the winter. The humidity levels should be kept above 50%.

– A pool with fresh drinking water should be available at all times. If this is not possible, drinking water receptacles must be secured so that they cannot be overturned. If there is no outdoor bathing area and the animal(s) must be kept inside for long period of time, a pool is highly recommended. Pools should be large enough for two adult tapirs to completely submerge their bodies. Safe and easy entrance/exit to the pool should be provided by gradual inclines and non-skid surfaces. Tapirs can hold their breath underwater for as long as 2-3 minutes. Swimming stimulates digestion in tapirs, and tapirs frequently defecate in their pools. The absence of pools or bathing areas may hinder proper digestion and can increase the incidence of rectal prelapse.

– Indoor areas should be cleaned daily and disinfected at least once a week. The bedding materials (grass hay or pine shavings) should be remove daily. All pools should be dumped and re-filled daily unless a filter system is present.

– Depending of the species, indoor facilities should enable the keepers to work with animals in a protected environment. This is especially recommended for Malayan tapirs.

– The roof should not be too translucent; excessive contact with sunlight can result in eye problems.

– As tapirs are very sensitive to lung infections, the indoors facilities need to be well ventilated. In the case of cold weather zoos, the exposure to high indoor temperatures and low outdoor temperatures within short intervals of time should be avoided.

– Tapirs require enough space for exercise, breeding, training sessions, and for veterinarians to conduct medical procedures. Enclosures should provide at least 60 m2 (645 ft2) for each animal. Visitors should be kept a minimum of 90 cm (3ft) away from tapirs. Visual barriers within outdoor enclosures are encouraged to allow animals to voluntarily isolate themselves from one another. Often subordinate animals separate themselves from a dominant animal, and females seek isolation prior to parturition.

– Tapirs can be easily maintained in shallow dry slanted moats with a 2m (6 ft) vertical outer moat wall. Enclosures without moats should have a minimum of 2m (6 ft) high barriers. Tapirs do not jump up but can easily climb over vertical walls as high as 1,20m (4 ft).

– Tapirs need shade in their outdoor exhibits at all times of the day, mainly in summer season and in zoos in hotter climates. Trees, forest vegetation, and artificial roofs, can all be used to create shade within outdoor enclosures. If an enclosure does not have adequate shade, during the hottest hours of the day tapirs should be moved into indoor facilities with lower temperatures and adequate shade.

– Outdoor exhibits should have soft substrates such as tilled earth. Sand can be used but should be avoided if possible. Ingesting sand can cause digestive problems in tapirs such as colic, intestinal impactions or intestinal obstruction. Tapirs should not be kept on concrete surfaces in order to prevent chronic foot problems and lameness.

– It is very important that the outdoor enclosure has a pool where tapirs can swim and defecate. Access to a pool or a natural pond is particularly important in warm climates. The pool should be cleaned and refilled with fresh water daily. Prior to and during cleaning, animals should be transferred to an adjacent pen. As mentioned above, when tapirs don't have access to a pool they are at a higher risk of experiencing rectal prolapses.

– When a tapir female gives birth and has access to outdoor exhibit, the pool must be empty to ensure that the baby does not fall into the water following his/her mother. This is essential in cold climate zoos, where there are reports of baby tapirs dying of pneumonia after entering pools during the winter.

– Environmental enrichment is another important consideration to improve the quality of life of captive tapirs. There is a wealth of information about this subject available online and in print. The following are important resources for tapir enrichment ideas: Tapir Specialist Group (www.tapirs.org); AZA (www.aza.org); Environmental Enrichment for All Species of Tapir in Captivity (Compiled by Maria Elisa Hobbelink); Enrichement for Tapirs (Graziele Moraes and Eliana Ferraz) in The Shape of Enrichment – Vol 18, Nº 1&2. 2009.

Tapirs are primarily browsing generalist herbivores that ingest several parts of plants, including leaves, fruits and tender stems, as well as grasses, shrubs, fruits, and twigs (Medici et al., 2007; Medici, 2010; Medici, 2011). They naturally consume multiple small meals throughout the day considering their limited stomach capacity. This should be taken into account in captivity where the total amount of daily intake for a mature adult tapir should be approximately 4% to 5% of the tapir's total body weight spread out over two to three meals (Janssen, 2003; Díz 2006). The food should be cut up into bite size pieces and fed fresh each day, food should be placed in separate cement containers afor each speciment. Food containers must be cleaned daily.

The consumption of foods with a high level of hydrolysable carbohydrates such as sugar, and/or rapidly fermentable carbohydrates (pectins) may result in abnormal fermentation in the hindgut. Such foods, combined with feeding schedules inconsistent with the species needs can contribute to colic, torsion or other digestive related pathologies. Diets with small quantities of fiber can lead to rectal prolapse.

As a ceco-colinic hindgut fermenter, the tapir’s gastrointestinal tract is very similar to that of the horse (Equus caballus). Four fibrous teniae create sacculations in the cecum and the colon is enlarged and attached to the cecum by fibrous tissue.The gall bladder is absent, but the bile duct empties into the duodenum 7.5 cm (2.95 in) from the pylorus.

In general, it is recommended that the diet is based mainly around a high quality commercial concentrate for horses, alfalfa and foliage. Fruits and vegetables should be present, but in smaller amounts. Mineral and vitamin supplementation is important.
Again, adequate fiber is very important in tapir diets. They should also consume some fruits and vegetables, but in smaller amounts; green leaves; tender stems; and nutritionally balanced food concentrates; among others items.

Diets should ideally be developed according the recommendations of nutritionists. It is important that zoos and other institutions with captive tapirs heed the advice from different Nutrition Specialist Groups. Important sources of information include the Nutrition Scientific Advisory Group (NAG) feeding guidelines (http://www.aza.org/nutrition-advisory-group/); veterinarians and veterinary committees; AZA Taxon Advisory Groups (TAGs); and Species Survival Plans® (SSP) Programs. The Tapir Specialist Group has a Tapir Nutrition Advisor, Dra. Julieta Olocco Díz (E-mail: mjoloccodiz@yahoo.com.ar).

12.4 Loading and Transport

Recommended protocol for loading and transport of tapirs in captivity is available in Chapter 4. Chemical Restraint.

12.5 Hand rearing orphan tapir calves

At times, in the case of wildlife rescues or unexpected deaths in zoos, it may be necessary to hand rear an orphan tapir calf. The orphan calf should be fed a formula that resembles the nutritional profile of tapir milk. If available in a sanitary environment, tapir calves can consume goat milk or cow's milk. In either case, these milks from other species should be diluted and supplemented to avoid constipation in the tapir calf. The follow is an example of replacement formula using cow's milk (Olocco 2004, personal communication):

- Cow whole powder milk: 100 gr.

- No-lactose powder milk: 50 gr.

- Skimmed powder milk: 50 gr.

- Water: 800 gr.

- Vit E + Selenium (according to specification of the laboratory)

It is important that the ingredients are mixed according to the recommendations of the product and/or the laboratory. The temperature of the formula should be about 36ºC in order to be sure the acceptance of the formula.

During the first 4 days of the life, the newborn has to feed every 2 hours during the day, with two fasting periods of 4 hours during the night. After 5 day of life, the frequency of feeding can be reduced to every 4 hours during the day, with a fasting period of 6 hours during the night.

In those tapirs that develop normally, up to the 10th day of life they can be feed every 6 hours during the day, with a fasting period of 8 - 10 hours during the night. As the calf continues to develop, the frequency of feeding can be incrementally decreased, but it is important to increase the amount of formula at each meal to ensure the replacement diet meets the animal's nutritional requirements.

In the first days of life, the newborn must consume about 10 to 15% of its body weight in milk per day. This quantity should be increased daily. By the 7th day after birth, the calf should consume approximately 20-28% of its weight in milk per day. The daily increase of milk quantity must be gradual to avoid abdominal distensions, enteritis, constipation or diarrhea. Between the 8th and 10th days of life, some leaves of alfalfa can be introduced to the baby's diet; in the wild the tapir's baby start feeding on solid food during the first week after birth (Olocco 2004, personal communication).

It is necessary to record the body weight of the tapir calf on a daily basis; healthy tapir calves increase in weight between 350 to 500 g per day (Quse et al. 2004).

After each feeding session it is important to massage the ano-genital area in order to stimulate the defecation and urination of the baby.

RECOMMENDED LITERATURE

AZA Tapir TAG. 2013. Tapir (Tapiridae) Care Manual. Association of Zoos and Aquariums, Silver Spring, MD. p. 65.

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Medici EP. 2010. Assessing the Viability of Lowland Tapir Populations in a Fragmented Landscape. PhD Dissertation. Durrell Institute of Conservation and Ecology (DICE), University of Kent. Canterbury, UK.

Medici EP. 2011. Family Tapiridae (TAPIRS). In: DE Wilson & RA Mittermeier (Eds.). Handbook of the Mammals of the World - Volume 2: Hoofed Mammals. Lynx Edicions, Spain.

Shoemaker, A. H. Barongi, R., Flanagan, J., Janssen D., Hernandez-Divers S.2003. Husbandry Guidelines for Keeping Tapirs in Captivity. Tapir Specialist Group; www.tapirs.org/Downloads/standards/tapir-TAG-husband-stan-en.doc.

Picture: Daniel Zupanc

13

Treatment and Management Protocol

Treatment and Management Protocol

The TSG Veterinary Committee has developed treatment protocols for some of the most common diseases in tapirs in order to be able to respond efficiently to the people who consult us. Dr. Donald Janssen made substantial contributions to these protocols based on his extensive field veterinary experience. He has improved and greatly enriched this protocol.

This manual includes treatment protocols for the following diseases:

1. - Hemorrhagic, Vesicular and Pustular Skin Disease

2. - Corneal Cloudiness

3. - Respiratory Diseases

4. - Lameness

5. - Colic

6. - Mandibular Abscessation

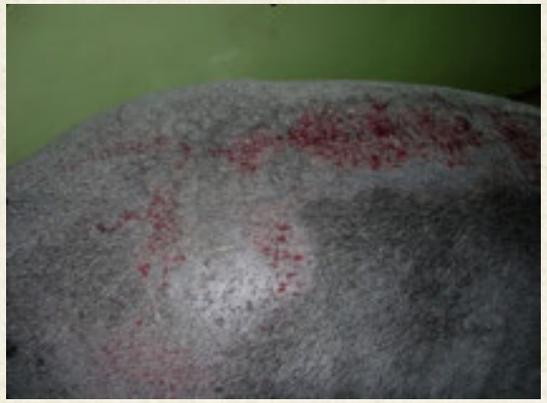
13.1 Hemorrhagic, vesicular and pustular Skin Disease

Skin diseases are very common in captive tapirs, although they have also been observed in the wild. The specific cause of dermatitis is unknown but may be related to bacterial infections (eg. Staphylococcus, Streptococcus); fungi (eg. Microsporum sp.; Trichophyton sp.); mites (eg. Sarcoptes); viruses; nutritional deficiencies; low serological concentrations of Copper (Vercammen et al. 2003); dirty enclosures; excessive sunlight (photosensitization); lack of access to water where tapirs can bath, among other causes.

Skin lesions occur mainly on the back and lumbo-sacra body regions. Sometimes dermatitis clears up without treatment, but in most cases it is necessary to treat affected individuals. Vesicular Disease Skin (Finnegan et al., 1993) is characterized by the presence of papules and vesicles with sero-sanguinous liquid. Severe cases may be associated neurologic signs characterized by ataxia, prostration and laminitis. Ramsay & Zainuddin (1993) hypothesize that Pox virus infection may be responsible for the formation of vesicles and pustules in some cases.

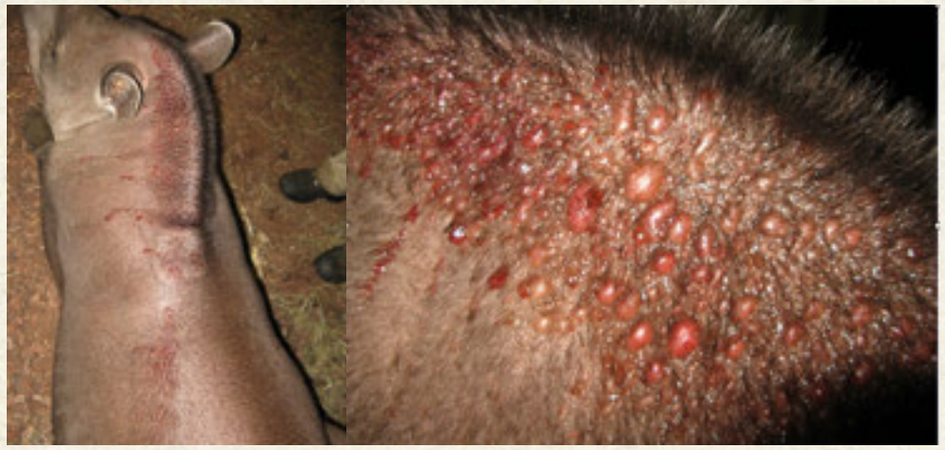
The characteristics of skin lesions on tapirs in captivity should be evaluated (hemorrhagic secretions, presence of vesicles or pustules). After this initial evaluation, it is necessary to observe the environmental conditions and review the diet of tapirs to determine if the condition of the enclosure or nutritional deficiencies may be causing the skin disease.

Gallery 14 - Skin Disease



Dorsal skin bleeding in Tapirus indicus in captivity. Photo: Zainal Zahari Zainuddin.

Desquamation and alopecia in Tapirus terrestris in captivity. Photo: Temaiken Foundation.



Hemorrhagic pustular dermatitis. Photo: Dorothée Ordonneau

Purulent dermatitis in Tapirus terrestris. Estación de Fauna Autóctona de Salta. Photo: Ricardo Bastida.

Prior to the treatment, it is important to carry out the following:

Clinical Tapir Management:

1) Clean the affected area:

a) Clean and disinfect the affected surrounding areas with antiseptics such as: Chlorhexidine 4% or 5%; Iodopovidona (Betadine®, Pervinox®); Chloroxylenol (Espadol®), or similar products.

b) In cases where a sample must be taken for a bacteriology study, it is recommended to clean the skin with Physiological Solution, not antiseptics.

2) Blood analyses

a) Perform blood analysis in order to assess the general health condition of the tapir. Evaluate the serum or plasma concentrations of copper, iron, zinc, beyond routinezserological determinations.

3) Sampling (use sterile gloves to protect your hands):

a) Samples for bacteriology study should be taken once the skin is properly cleaned with sterile Physiological Solution. After this, gently rub a sterile swab on affected area (bleeding, discharge sero-sanguinous fluid, pustules, etc.) and place the swab in a transport medium or in a nutrient culture medium (eg Stuart) or another medium recommended by the laboratory. If pustules with serosanguineous fluid are present, take a sample using a sterile needle and syringe, close tightly and send to the laboratory. Label each sample and keep refrigerated. Request an anantibiogram.

b) Samples for mycological study should be taken once the skin is properly cleaned with sterile Physiological Solution or alcohol 70%. With a sterile scalpel blade make a scraping of the affected area (usually hairless areas) and its edges. Place the sample in a sterile tube with transport medium or in sterile Petri dish(could use some medium recommended by the laboratory). Remove several hairs from the edges of the affected area and in other parts of the body where lesions are observed. Place the hairs in a sterile tube or sterile Petri dish. Label the samples and store them at room temperature. Request an antimicogram.

c) Perform a cytology study from affected areas.

d) Virological study: skin biopsies or swabs from the lesions should be frozen and samples analyzed using PCR when possible.

e) A Histopathology study is useful for evaluating the tissue conditions. Skinbiopsies from both affected and healthy areas are recommended to allow the pathologist to compare the microscopic characteristics of the different skin tissues.

Size of the sample: Depending on the affected area, a 0.5 X 0.5 X 0.5 cm sample is ideal to ensure the penetration of the fixative. The samples should be handled carefully using a dissection clamp without teeth to prevent tissue damage. The samples should be placed in 10% buffered Formalin, or Bouin solution. Label the samples and send it to the laboratory.

f) Evaluate the presence of ectoparasites. In this case, take hair samples (even with bulbs) from affected body area. Complement hair samples with samples of skin scrapings from different parts of the body. Place the material in a Petri dish or sterile tube. Label the samples and send it to the laboratory.

g) As mentioned above, it is important to evaluate the diet of affected tapirs and determine levels of Copper, vitamins, minerals, etc. A serological test for copper is also important.

h) Again, an evaluation of the environmental conditions including the sanitary conditions of the enclosure, shade levels and the presence/absence of clean water in which tapirs can bathe, may help diagnose the cause of skin diseases. For more information consult the Husbandry Manual for Tapirs in Captivity in www.tapirs.org

Examples of treatment (depending the diagnosis):

If the dermatitis is caused by bacteria/s, we recommend the following protocol:

- Wash the affected areas with Chlorhexidine or other antiseptic (twice a day).

- Apply a dermal cream bactericide on a daily basis (take in account the result of antibiogram). Apply the cream after disinfecting of the affected areas (twice a day).

- If necessary, give an oral antibiotic.

- Improve the hygiene of the enclosure, food and water containers, pool water, etc.

- If necessary, separate the sick tapir from other tapirs in order to prevent further transmission.

- Repeat bacteriological culture 14-21 days after the start the treatment to evaluate its efficacy.

If the dermatitis is caused by dermatophytosis, we recommend the following:

- Wash the affected areas with Iodopovidona (twice a day).

- Administer Griseofulvin (10mg/kg VO) for 60 days.

- Apply an antimicotic skin cream on a daily basis (take into account the result of antimicogram). Apply the cream after disinfecting of the affected areas.

- Evaluate hemogram and serum enzymes (eg hepatic enzymes) during the treatment.

- Maintain good hygiene of the enclosure, food and water containers, pool water, etc.

If the dermatitis is caused by environmental factors (excessive sunlight, poor sanitary conditions in the enclosure, among other factors), we recommend the following:

- Wash the affected area with an antiseptic such as Chlorhexidine (twice a day).

- Daily application of sunscreen cream (2-3 times daily).

- Increase shade in the outdoor enclosure with more vegetation (shrubs, trees) or artificial materials.

- Provide a pool to allow tapirs to bathe daily.

- Maintain a good hygiene of the enclosure and the rest area.

- Apply mosquito repellent spray around the lesions.

- Keep the tapir in a shaded area (avoid outside the enclosure) until skin conditions improve.

In case of nutrient deficiency in the diet, we recommend the following:

- Work with a nutritionist to formulate an adequate diet with appropriate concentration of Aminoacids, Biotin, Vitamins and Copper. Supplement with a multivitamin-mineral for horses.

RECOMMENDED LITERATURE

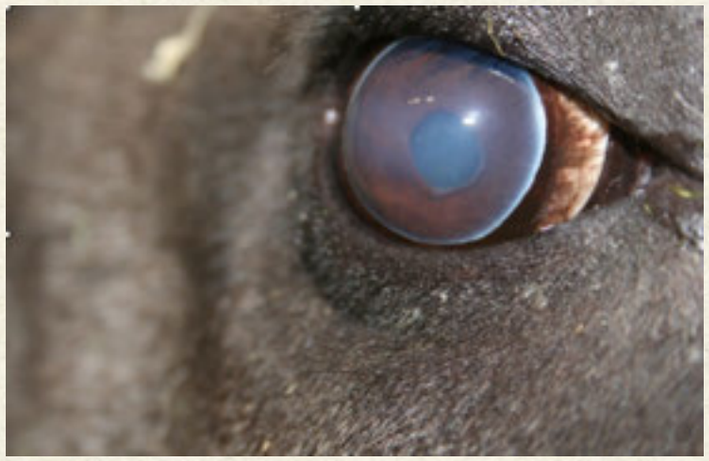
Finnegan M, Munson L, Barrett S and Calle P. 1993. Vesicular Skin Disease of Tapirs.AAZV Conference (Abstracts).

Ramsay E & Zainuddin Z. 1993. Infectious diseases of the rhinoceros and tapir.Journal of Zoo and Wildlife Medicine, 3, 459-466.

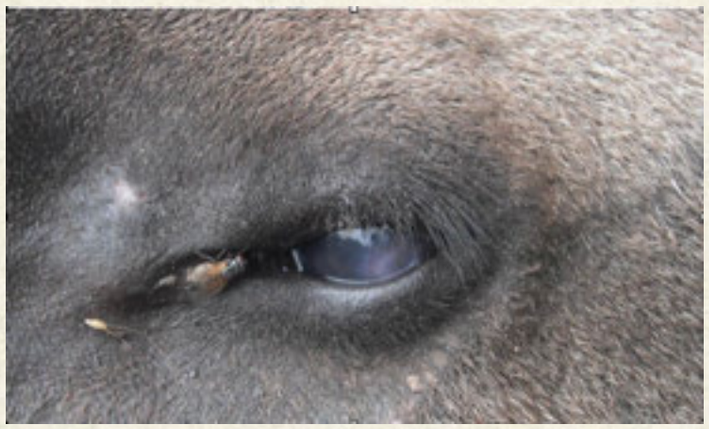
Vercammen F, De Deken R and Brandt J. 2003. Dorsal Skin Bleeding in a MalayanTapir (Tapirus indicus). Verh.ber. Erkrg. Zootiere, 41.

Corneal cloudiness is very common and frequent in tapirs. The specific cause is unknown but is probably a result of trauma, bacterial or viral infections, high exposure to sunlight, or a combination of these factors. Occasionally corneal cloudiness is associated with corneal ulceration or alterations of the deeper structures of the eye.

Gallery 15 - Keratoconjuntivitis



Corneal cloudiness, following a chronical keratoconjunctivitis. Photo: Dorothée Ordonneau.



Keratoconjuntivitis in Tapirus terrestris in captivity. Photo: Soledad de Bustos (Estación de Fauna Autóctona de Salta (Argentina).

As with most other conditions to which tapirs are particularly susceptible, it is recommended to treat both the tapir and address issues in their environment (captivity). It is recommended to carry out an eye examination to determine the type of injury and which ocular structures are involved. We recommend the following management steps before deciding on a specific treatment:

Clinical Tapir Management

a) Evaluate the presence of wounds or other alterations in the structures surrounding the eye: eyelids, conjunctiva, lachrymal glands and eyelashes.

b) Take a sample with a sterile swab for bacterial culture and antibiogram. If possible, conduct viral research, such as tests for herpes virus.

c) In trained or anesthetized tapirs, an ultrasound exam is recommended to evaluate ocular deep tissue injuries, the presence of foreign bodies in the eye, lens dislocation, etc.

d) In order to diagnose a corneal ulceration, it is necessary to apply a few drops of Fluorescein (solution of 0.5% to 2%), which impregnates the green color of the injured tissues. After this, it is necessary to wash the eye with sterile Physiologic Solution. When ulceration is present, the green color will persist.

Environmental Management (in captivity):

a) Provide additional shade in the enclosure to reduce the excessive exposure light. Different options for achieving this are describe above, but include increasing natural vegetation in the enclosure or adding artificial sources of shade.

b) Ensure that the enclosure has a good water source (pool, lake, etc.) where tapirs can dive and swim. For more information regarding the necessary characteristics of the tapir´s environment see Chapter 12 of this Manual or consult TSG Husbandry Manual for Tapirs in Captivity in www.tapirs.org

c) Minimize the presence dust, dirt, smoke or any pollution that could damage the eyes of tapirs inside their enclosures.

Examples of Treatments:

Example 1:

-Wash the eyes with sterile Physiologic Solution (4-5 times per day).

- Apply antibiotic eye drops (eg Tobramycin; Netilmicin; Chloramphenicol, Neomycin) 4-5 times a day and after the eyewash. Take into account the antibiogram result.

- Ensure the enclosures have good shade and hygiene.

- Use protective ophthalmic ointment as needed (eg. Dimeticone, carbomere).

Example 2:

-Wash the eyes with sterile Physiologic Solution (4-5 times per day).

- Apply antibiotic eye drops (eg Tobramycin; Netilmicin; Chloramphenicol, Neomycin) 4-5 times a day and after the eyewash. Take into account the antibiogram result.

- When there is no corneal ulceration apply antibiotics in association with corticosteroids.

- Good shade and hygiene in the enclosure.

- Ensure the enclosures have good shade and hygiene.

- Use protective ophthalmic ointment as needed (eg. Dimeticone, carbomere).

Tapirs are very susceptible to respiratory diseases. They are commonly the result of bacterial infections caused by species of the following genera: Streptococcus, Klebsiella, Corynebacterium, Mycobacterium and Fusobacterium. Some mycoses can also cause respiratory problems; there are some reports about respiratory illnesses caused by fungal Coccidioidomycosis that have resulted in deaths. Viral and parasitic infections can also be the causes of respiratory diseases.

In captivity in Europe, lowland tapirs quite often exhibit nasal discharge without any pulmonary infection. This is likely primarily due to their environment (sand, dust, etc.) (Dorothée Ordonneau, personal communication).

When tapir present symptoms such as dyspnea, nose secretion, cough, respiratory, rales, anorexia, or fever, it is recommended to make a good diagnosis based on a thorough clinical examination of the specimen accompanied by complementary observations and tests. It is important to differentiate between diseases of the upper and lower airways.

Clinical Tapir Management

We recommend the following protocol in cases of respiratory illness.

a) Perform blood analysis (hemogram and serology).

b) As needed, take chest X-Rays in order to evaluate the lung condition. Portable X-Ray equipment may be adequate.

c) Collect samples with respiratory secretions for bacteriology and mycology diagnostic tests. Request antibiogram and antimicogram.

d) If necessary, anesthetize tapirs and perform tracheal washings with sterile Physiologic Solution in order to sample deeper airways. If there is no experience with this technique, request the assistance of a trained veterinary specialist with experience performing tracheal washings in horses. For anesthesia consult the Chapter 4 (Chemical Restraint) in this Veterinary Manual.

e) Take blood samples when fever is particularly acute and perform bloodculture (follow the recommendations of the lab).

f) Develop a spreadsheet with baseline information on respiratory rate, and note important breathing characteristics: superficial, deep, apneas, etc. Record the body temperature in the same spreadsheet.

g) Keep the tapir isolated to prevent infections to other tapirs.

Examples of Treatments:

Example 1(if the respiratory disease was caused by bacteria/s):

- Provide antibiotics specific to the bacteria isolated.

- In cases where bacterial isolation and antibiogram were not possible, apply a broad-spectrum antibiotic.

- Evaluate the hydration of the tapir. If necessary, hydrate with a Dextrose Solution or another commonly used solution, depending on the serological results.

- Administer Vitamin complex.

- Improve hygiene of the enclosure and the food and water containers.

- Perform new blood analysis 48-72h after initiation of treatment.

-The use of bronchodilator such as Clenbuterol could relieve the animals during the antibiotic treatment.

Example 2 (if the respiratory disease was caused by fungal infection):

- Provide a broad antifungal, such as Itraconazole, or another antimicotic indicated by the laboratory according to antimicogram. Take into account that long term antimicotic treatment could lead to severe anemia, for this reason will be necessary the control of hemoglobin concentration regularly throughout treatment.

- Evaluate the hydration of the tapir. If necessary, hydrate with a Dextrose Solution or another commonly used solution, depending on the serological results.

- Administer Vitamin complex.

- Improve hygiene of the enclosure and the food and water containers.

Management and treatment examples in case of Tuberculosis suspicion:

Tapirs are very susceptible to Tuberculosis (TB), and it is the most common pulmonary illness. In tapirs in captivity veterinarians have isolated Mycobacterium bovis, responsible for causing tuberculosis in cattle and humans; and Mycobacterium pinnipedii, responsible for causing TB in pinnipeds (Cousins et al., 2003; Bastida et al., 1999; Bastida etal., 2011; Hoyer, 2011; Jurczynskiet al. 2011).

If the veterinarian suspects that captive tapirs are infected with TB, perform a thorough examination of tapir including a blood analysis in order to evaluate the general health condition; sample nasal or respiratory tract discharge to carry out the necessary bacterial culture. To complement these exams, it is necessary to conduct an ELISA test; DNA tests, and other analyses.

If TB is confirmed, the personnel in charge of the tapirs will have to decide if it is necessry to euthanize the tapir(s). In cases where treatment is a viable option, it is necessary to admnister a suite of antibiotics such as Isoniazid, Rimfampicina, Ethambutol, Pyrazinamide and Rifabutin. The treatment protocol will also require several months of regular clinical exams.

RECOMMENDED LITERATURE

Cousins D, Bastida R, Cataldi A, Quse V, Redrobe S, Dow S, Duignan P, Murray A, DupontC, Ahmed N, Collins D, Butler W, Dawson D, Rodríguez D, Loureiro J, Romano MI, Alito A,Zumárraga M, Bernardelli A. 2003. Tuberculosis in seals caused by a novel member of theMycobacterium tuberculosis complex: Mycobacterium pinnipedii sp.nov. International Journalof Systematic and Evolutionary Microbiology 53, 1305-1314.

Bastida R, J Loureiro, V Quse, A Bernardelli, D Rodríguez, E Costa. 1999.Tuberculosis in aWild Subantarctic Fur Seal from Argentina. Journal of Wildlife Diseases35(4): 796-798.

Bastida R, Quse V, Guichón R.2011.La tuberculosis en grupos de cazadores recolectoresde Patagonia y Tierra del Fuego: Nuevas alternativas de contagio a través de la fauna silvestre.Revista Argentina de Antropología Biológica, 13,1.

Hoyer M. 2011.Management of a TB Positive Malayan Tapir (Tapirus indicus) Breeding Couple under Zoo Conditions. In: Procceding Fifth International Tapir Symposium p31.

Jurczynski K; Lyashchenko KP; Gomis D; Moser I; Greenwald R; Moisson P. 2011.Pinniped tuberculosis in Malayan tapirs (Tapirus indicus) and its transmission to other terrestrial mammals.In: Journal of Zoo and Wildlife Medicine42(2):222-7.

Lameness (inflammation of the hoof corium) is very common in tapirs; the most common cause is frequent walking on a hard and abrasive substrate, sudden activity due to enclosure changes or introduction of conspecifics. Dirty substrate can also contribute to this affection. Working to prevent foot injury is much more effective and less of a hassle han waiting until it is necessary to treat lameness.

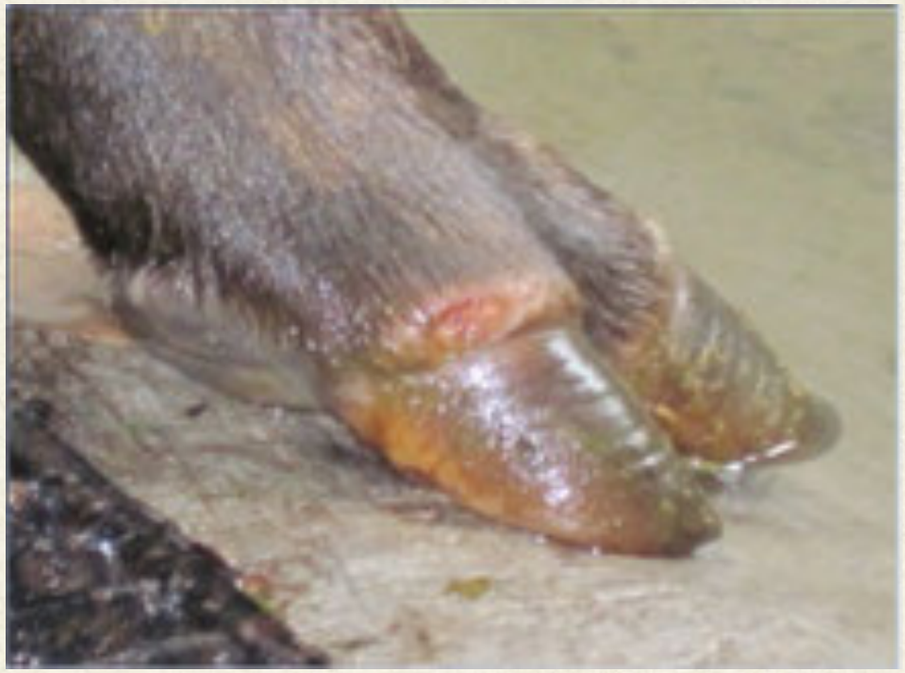


Figure 51 - Hoof infection in Tapirus bairdii. Photo: Jonathan Perez.

At times there can be significant tissue damage, and sometimes sole ulcerations are present. Common clinical signs include: tapirs walk with difficulty, there is pain and different claudication degree, animals refuse veterinary examination due the pain, and animals decrease food intake.

As with prior diseases and conditions, is important to treat the individual and evaluate and correct the enclosure and substrate characteristics that could be causing problems. In some cases, a simple change to the tapir enclosure, such as adding a soft substrate, can reolve the problem within a few weeks without treatment. Obesity can exacerbate the lesions and slow the recovery. If obesity is the suspected cause, a re-evaluation of the tapir diet will be necessary.

Examples of treatments:

Example 1:

- Administer Flunixin Meglumine (Banamine): 1.1 mg/kg every 24h (IM) (equine dose). Repeat the dose the next day if symptoms persist. It can also be given orally. Do not exceed a week of treatment.

- Administer Phenylbutazone (equine dose), or meloxicam (equine dose) orally.

- Administer antibiotics such as Tylosin; Penicillin Streptomycin; TMP/sulfamids, Enrofloxacine (1 mg/kg) orally.

- Place the tapir in a soft and clean substrate.

- Evaluate the diet (in horses the excess of carbohydrates predisposes to lameness).

Example 2:

- Administer Phenylbutazone: 4 to 8mg/kg/24h (VO or IM) (in horses the dose may be2.2mg/kg/12h or 4.4 mg/kg/24h VE).

- Administer Antibiotics such as Tylosin or Penicillin Streptomycin.

- Place the tapir in a soft and clean substrate.

- Evaluate the diet (in horses the excess of carbohydrates predisposes lameness).

Example 3:

- Administer Flunixin Meglumine (Banamine): 1.1mg/kg/24h (IM). Repeat the dose the next day if symptoms persist.

- Administer Tramadol (2mg/kg) PO c/12h X3 days.

- Administer Trimethoprim sulfa (30mg/kg) PO every 24h X4days.

When laminitis is accompanied by cracks of the hoof, cleaning and bandage the feet is necessary. Once per day, the veterinarian can apply Hooflex oinment (Cloroxylenol) once a day.

As in other Perissodactyla –like horses- tapirs are very susceptible to colic. It can have several different causes, but is most commonly caused by problems with the diet: such as diets with high or low fiber; innappropriate feeding schedules, including giving tapirs a high volume of food offered once a day (due to the reduced size of th estomach, tapirs should eat small amounts of food several times a day).

Ingestion of soil, sand or other foreign material can also cause colic and has even resulted in the death of captive tapirs. Colic can also be caused by bacterial enterocolitis, intestinal disorders such as volvulus, intestinal torsion, and obstructions, among other problems.

Clinical Tapir Management

a) Evaluate the hydration status of the tapir.

b) Perform blood analysis.

c) Use contrast radiography in order to detect the presence of possible foreign bodies in the GI tract.

d) If appropriate, perform a copro-culture and antibiogram. As bacterial flora is normally present in the intestine, it is recommended that the veterinarian request what species of bacteria should be isolated and could possibly be causing colic (eg.Escherichia coli, Salmonella). This information will help to guide the laboratory.

e) Determine the presence of diarrhea and its characteristics.

Examples of Treatments:

a) In most cases treating symptoms is sufficient. The precise treatment varies based on the cause of colic.

b) If the tapir has diarrhea, provide electrolytes intravenously or administer oral hydration salts in order to replace fluids lost during the diarrhea.

c) When colic is due to a high presence of parasites, deworming should be done for the parasite/s identified. The antiparasitic drugs and dosage are similar to those used in horses: Thiabendazole (44mg/kg VO); Mebendazole (8mg/kg VO); Cambendazole (20mg/kgVO); Tetramisole (9mg/kg VO).

d) If colic is due to a diet with low or high quantity of fiber, a nutritionist must be consulted to correct the nutritional issue.

e) If pain is severe, adminster: Flunixin Meglumine (Banamine: 1.1 mg/kg every 24 h) (IM). If symptoms persist, repeat the next day.

f) Evaluate if the tapir can urinate and, if possible, evaluate the characteristic of the urine.

g) Do not provide food during the first 24 hours of treatment. Once colic symptoms decrease, gradually incorporate good quality food according the recommendations of nutritionist.

h) Emergency surgery can be necessary for torsion, volvulus, or obstruction. Consult an equine surgeon for diagnostic indications and surgical options.

13.6 Mandibular Abscessation

The mandibular abscessation ocurrs very frequently in tapirs and could be the result of injuries in the mouth becoming infected by different bacterias such as Corynebacteriumpyogenes; Streptococcus sp., Actinomyces sp. y Necrobacillus sp. Periodontal diseases, molar apical abscesses or gingival infections are other causes of this disease. It can become chronic and result in osteomyelitis, which can result in systemic infection and the death of the tapir.

Clinical Management of tapir:

a) Use sterile gloves in order to protect hands from mucous.

b) With a sterile swab, take samples of the content of the abscess (if it can be drained); place the swab in a transport medium or in a nutrient culture medium (eg. Stuart), or another medium recommended by the laboratory. Label the simple and keep refrigerated until it is sent to the laboratory. Request an antibiogram.

c) In case that the abscess cannot be drained, it will be necessary to anesthetize the tapir and take samples from the internal wall of the abscess. Place the sample in a transport medium or in a nutrient cultura medium (eg. Stuart), or another medium recommended by the laboratory. Label the sample and keep refrigerated until it can be sent to the laboratory. Request an antibiogram. Consult Chapter 4 (Chemical Restraint) in this Veterinary Manual for recommended anesthesia protocols.

d) If necessary, take X-Rays of the affected area in order to evaluate the condition of the bone and the teeth.

e) If it is necessary to anesthetize tapirs, we recommend performing a biopsy of the lesions and the surrounding areas.

f) Perform blood analysis to evaluate hemogram, eritro sedimentation and serological enzymes concentration.

Examples of Treatments:

a) Under sedation or anesthesia of the tapir, drain abscess content and clean its cavity with Clorhexidine. Remove the damage tissues and purulent material.

b) Perform surgical debridement of the affected bone, teeth or necrotic tissues.

c) Give broad-spectrum antibiotic over an extended period of time. Take into account the antibiogram results when choosing antibiotic.

d) In the case of intensive pain, administer Flunixin Meglumine (Banamine), 1.1mg/kg/24h (IM).

e) Feed the affected tapir a diet of soft foods (fruits, fresh leaves, etc.) to avoid lesions in the oral cavity during the treatment.

f) Ensure that the water and food containers are properly sanitized. Ensure that the enclosure is kept in good, sanitary conditions and includes no abrasive materials.

Picture: Byron Jorjorian

APPENDIX

APPENDIX 1

Agents Commonly Used for the Chemical Restraint of Tapirs

Alpha-2-Agonists: Medetomidine, Detomidine, Romifidine, Xylazine / Reversal Drugs: Atipamezole, Tolazoline

These drugs produce depression of the Central Nervous System (CNS), being classified as sedatives and soft analgesics, with myorelaxation properties. When using these drugs in tapirs it is necessary to consider their capability of depressing thermoregulation. In many species, these drugs produce emesis; however this does not seem common in tapirs. Typically these drugs cause an initial increase in blood pressure followed by a prolonged decrease. However, there are no specific studies on the effects these drugs have on the blood pressure of tapirs. Yet experience has shown that the drop in pressure can hinder blood collection from peripheral veins. It is possible to reverse the effects by administering atropine. Other circulatory effects include bradycardia and arrhythmias. Short apnea and exposure of the penis have also been reported as common with these drugs.The isolated use of Alpha-2-agonists has proven efficient during a series of chemical restraint procedures. In particular, Romifidine has shown the best results, due to the low volume required, low costs and predictable, stable effects on cardio respiratory parameters. In general, Alpha-2-agonists have been considered fundamental in the developing of simple and safe anesthetic protocols for tapirs. They have been successfully combined with dissociative drugs to produce deeper anesthesia both in field and captivity. They have also been combined with opioid derivates to produce safe chemical restraint and deep sedation for field capture and handling.

Opioid Derivates: Butorphanol Tartarate, Carfentanil, Etorphine / Reversal Drug: Naloxone, Naltrexone

The opioid derivates have been commonly used for the restraint and anesthesia of both wild and captive tapirs. They have been safely combined with Alpha-2-agonists and/or Ketamine to produce good analgesia and tend to have a stable, predictable effect on cardio respiratory parameters. The anesthetic recovery is typically fast and without complications. It is possible to let tapirs recover without a reversal agent or administer Naloxone.

Dissociative Drugs: Ketamine, Tiletamine / No specific reversal drugs

The dissociative drugs, derivates of ciclohexamine, may produce amnesia and catalepsy, providing an uncomfortable anesthetic induction and recovery, with ataxia, falls and pedaling movements (especially with Tiletamine = Telazol, Zoletil). Combinations of Tiletamine with Alpha-2-agonists in tapirs may produce periods of anesthetic respiratory depression. Sometimes the periods of apnea may require to be reversed by respiratory massage and respiratory stimulants. When Alpha-2 reversal agents are not used, the anesthetic recovery might be uncomfortable, with oscillations between consciousness and depression.

Atropine

In low doses, Atropine inhibits excessive salivation and respiratory secretions. In moderate doses, Atropine may be used to increase the hearth rate. Excessive doses, however, may reduce gastrointestinal and urinary motility. One of its most important uses in tapir anesthesia is to reduce hyper secretion and reverse the drop in blood pressure caused by to Alpha-2-agonists or dissociative drugs, which can hamper blood collection.

Emergency Drugs

During a chemical restraint's planning stages, it is highly recommended to estimate dosages of emergency drugs, so that these drugs are available and veterinarians are prepared to use them if needed. The use of Doxapram may be administered as a prophylactic in protocols using alpha-2-agonists, opioids or Telazol/Zoletil, to prevent respiratory depression.

APPENDIX 2

CHEMICAL RESTRAINT &CLINICAL EVALUATION

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IUCN/SSC Tapir Specialist Group (TSG)

CHEMICAL RESTRAINT & CLINICAL EVALUATION

Species: _____ Identification: _____

Sex: ( ) Male ( ) Female Age: _____ ESTIMATEDBODY WEIGHT: | REALBODY WEIGHT:

Location: _____ GPS: _____ Date: ___/___/_____

Team: _____

NutritionalCondition: ( ) Caquetic ( ) Lean ( ) Good ( ) Obese

Apparent Health: ( ) Good ( ) Regular ( ) Bad

Behavior: ( ) Depressed/apathy ( ) Calm ( ) Alert ( ) Excited ( ) Aggressive

Capture Method: ( ) Captivity ( ) Capture pen ( ) Pitfall ( ) Dart Shooting ( ) Hunting dogs

Weather: ( ) Dry ( ) Open ( ) Partially cloudy ( ) Cloudy ( ) Rainy

Temp. (°C): _____

Humid. (%): _____

Drug	Conc. (mg/ml)	Dose (ml)	DS	Via	Time
					:
					:
					:
					:
					:
					:

DS = Delivery System = DP (dart pistol), S (syringe), BP (blowpipe) etc. **Via** = SC, IM, IV etc.

Time	RR	RR type	HR	%O_2	T°C	CFT	RELAX	Comments
:								
:								
:								
:								
:								
:								
:								
:								
:								
:								
:								
:								
:								
:								
:								

RR = Respiratory rate (mpm), **HR** = Hearth rate (bpm), **%O_2** = blood oxygensaturation (%), **CFT** = Capillaryrefill time (s), **RELAX** = Muscular relaxing (none = 0, soft = 1, incomplete = 2, complete = 3), **RR type** = Respiratoryrate type (costal = C, abdominal = AB, costal/abdominal = CAB, superficial = S, deep = D).

GENERAL CLINICAL EXAM

Skin/Hair: ( ) Good ( ) Regular ( ) Bad **Comments:** _____

Ectoparasites: ( ) Few ( ) Regular ( ) Excessive **Which?** _____

Consolidated fractures: ( ) Yes ( ) No **Where?** _____

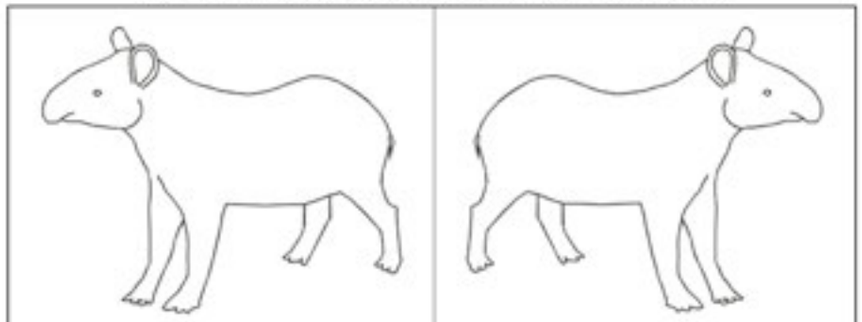
( ) **Mouth/Teeth/Nostrils:** _____

_____

( ) **Eyes/Ears:** _____

( ) **Vagina/Penis/Testicles:** _____

EXTERNAL LESIONS (cuts, abscesses, nodules, scars etc.)



LEFT RIGHT

Description of the lesions:

Identify and number the lesions on the drawings, and describe them in detail on the space provided above.

COLLECTION OF BIOLOGICAL SAMPLES

( ) Blood with anticoagulant, volume: _____ ml

( ) Blood without anticoagulant, volume: _____ ml

( ) Blood smears, no. of slides: _____

( ) Blood in absolute alcohol, no. of microtubes: _____

( ) Genetic samples, which?

_____

( ) Vaginal cytology, no. of plates: _____

( ) Ectoparasites, no. of flasks: _____

( ) Hair ( ) Feces

Swabs: ( ) Mouth ( ) Nostrils ( ) Eyes ( ) Ears ( ) Vagina ( ) Prepuce ( ) Rectum ( ) Lesions ( ) Eyes

Other samples (urine, milketc.):

Notes: _____

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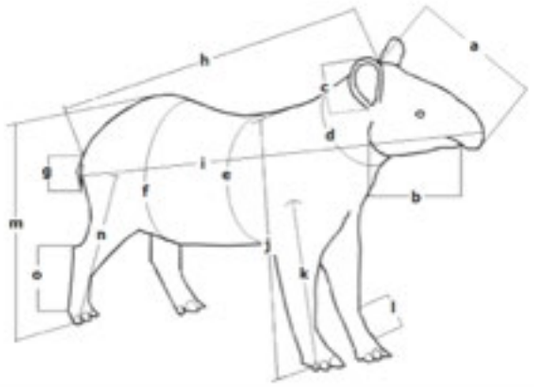
CORPORAL MEASUREMENTS

Species: _____ Identification: _____

Sex: ( ) Male ( ) Female Age: _____ Body weight: ____ ( ) ESTIMATED ( ) REAL

Location: _____ GPS: _____ Date: ___/___/_____

Photographs: ( ) FACE ( ) FULL BODY ( ) MOUTH/TEETH ( ) GENITALS ( ) PARTICULAR SIGNS



a. Head Length: _____	f. Abdomen Circ.: _____	k. Front Leg Length: _____
b. Jaw Length: _____	g. Tail Length: _____	l. Carpal Length: _____
c. Ear Length: _____	h. Trunk Length: _____	m. Rear Height: _____
d. Neck Circ.: _____	i. Full Length: _____	n. Rear Leg Length: _____
e. Thorax Circ.: _____	j. Front Height: _____	o. Tarsal Length: _____
Distance between eyes: _____	Left Testicle Length: _____	Right Testicle Length: _____
Vulva Length: _____	Left Testicle Circ.: _____	Right Testicle Circ.: _____

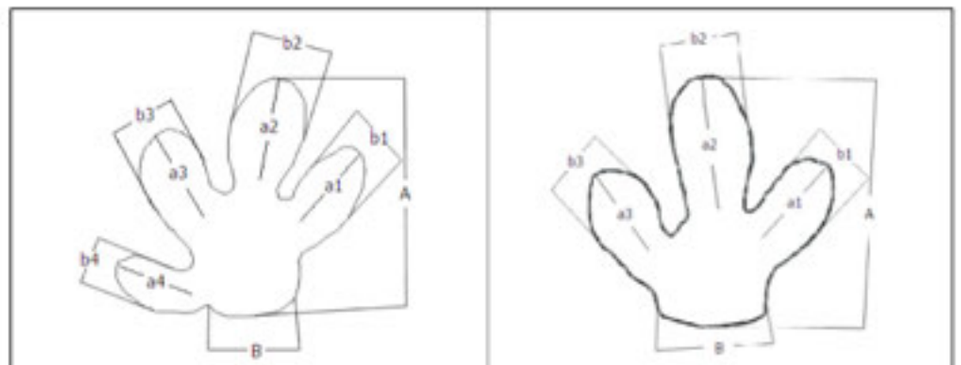
NOTE:All measurements should be taken with measure tape along the body, following its natural curves.

DENTITION (measurements and fractures)

Upper Left											Upper Right										
M3	M2	M1	P4	P3	P2	P1	C	I3	I2	I1	I1	I2	I3	C	P1	P2	P3	P4	M1	M2	M3
Lower Left											**Lower Right**										
M3	M2	M1	P3	P2	P1	C	I3	I2	I1		I1	I2	I3	C	P1	P2	P3	M1	M2	M3	

NOTE: Measure the length of incisors and canines, for other teeth mark a cross when a tooth is fractured or missing.

Notes:

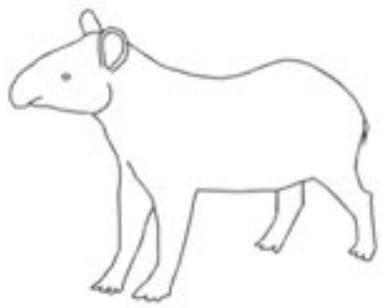


ANTERIOR FEET

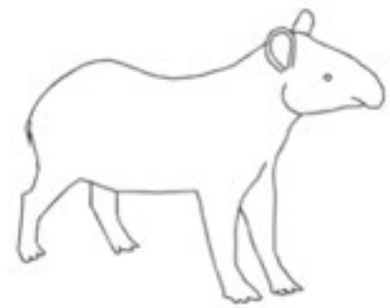
POSTERIOR FEET

Left Anterior Foot	a1: _____	a2: _____	a3: _____	a4: _____	A: _____
	b1: _____	b2: _____	b3: _____	b4: _____	B: _____
Right Anterior Foot	a1: _____	a2: _____	a3: _____	a4: _____	A: _____
	b1: _____	b2: _____	b3: _____	b4: _____	B: _____
Left Posterior Foot	a1: _____	a2: _____	a3: _____		A: _____
	b1: _____	b2: _____	b3: _____		B: _____
Right Posterior Foot	a1: _____	a2: _____	a3: _____		A: _____
	b1: _____	b2: _____	b3: _____		B: _____

NOTE: The digits are counted from the interior to the exterior of the feet.



LEFT



RIGHT

Description of the signs: _____

NOTE: Identify and number the particular signs on the drawings, and describe them in detail on the space provided above.

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NECROPSY

Performer of the necropsy:

Institution:

Address:

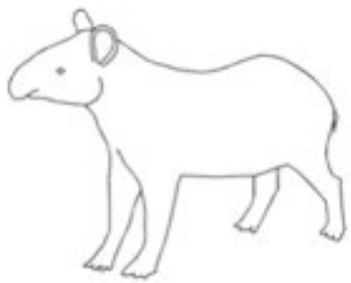
Species: Identification:

Sex: ( ) Male ( ) Female Age Class: Body weight: ( ) ESTIMATED ( ) REAL

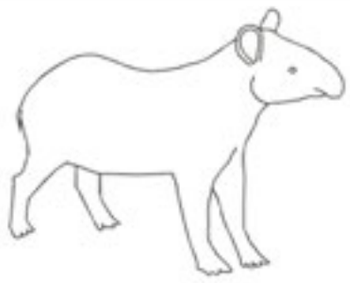
Dateof death (estimated): ___/___/_____ Dateof necropsy: ___/___/_____

Location: GPS coordinates:

Known history of animal / Circunstances of death:



LEFT



RIGHT

External exam (skin, scars, ectoparasites, natural orifices, nutritional condition):

Body cavities (peritoneum, pleura, pericardium, visceral positioning, cavitary liquids, fat stores):

Respiratory system (nasal cavity, pharynx, larynx, trachea, bronchi, lungs, regional lymph nodes):

Cardiovascular and hemolymphatic systems (heart, great vessels, spleen, lymph nodes, thymus):

Digestive system (mouth, teeth, tongue, esophagus, stomach, small intestine, cecum, large intestine, rectum, liver, pancreas, mesenteric lymph nodes):

Urinary system (kidneys, ureters, bladder, urethra):

Reproductive system (testes/ovaries, uterus & cervix, penis/vagina, urogenital canal, prostate, seminal vesicles, bulbo-urethral gland, mammary gland, placenta):

Nervous system and sensory organs (brain, meninges, spinal cord, peripheral nerves, eyes, ears):

Endocrine system (thyroids, parathyroid, adrenals, pituitary):

Musculoskeletal system (bones, bone marrow, joints, tendons, muscles):

Preliminary Diagnosis:

Samples collected for histopathology:

( ) Skin	( ) Trachea	( ) Lungs	( ) Myocardium
( ) Spleen	( ) Thymus	( ) Esophagus	( ) Stomach
( ) Small intestine	( ) Large intestine	( ) Cecum	( ) Liver
( ) Pancreas	( ) Kidneys	( ) Ureters	( ) Bladder
( ) Testes/Ovaries	( ) Male sexual glands	( ) Uterus	( ) Vagina
( ) Brain	( ) Meninges	( ) Spinal cord	( ) Pituitary
( ) Thyroids	( ) Adrenals	( ) Bone marrow	( ) Muscle

Other samples:

( ) Endoparasites	( ) Ectoparasites	( ) Stomach content	( ) Testes/Ovaries

( ) Toxicology: viscera, stomach content, hair, fat, cardiac blood, bone marrow. ( ) Genetic samples

( ) Microbiology (describe samples): _____

APPENDIX 3

Useful Websites Equipment and Supplies

Capchur

www.palmercap-chur.com

Dan-Inject

www.dan-inject.com

Pneu-Dart

www.pneudart.com

Dist-Inject

www.distinject.com

Followit

www.wildlife.followit.se

Telinject

www.telinject.com

Telonics

www.telonics.com

Reconyx

www.reconyx.com

Bushnell

www.bushnell.com