RESCUE AND REHABILITATION PROTOCOL FOR

JUVENILE/ADULT FRANCISCANA DOLPHINS

Pontoporia blainvillei

J. Meegan, F. Gomez, A. Barratclough, C. Smith, J. Sweeney,
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Alliance for Franciscana-Dolphin
Conservation Research, Rescue and Rehabilitation AFCR3

Integrated Conservation Planning for Cetaceans
IUCN SSC

AFCR3 - Publication Nr. 02 - 2023

Coordination

Partners

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This protocol was developed and implemented by veterinarians, technicians, biologists, and animal care and rehabilitation specialists. This protocol has been gradually applied for the past two stranding seasons. It is intended to be a living document, with modifications made as new knowledge is gained with each case, ultimately bringing us closer to the overall goal of successfully rehabilitating Franciscana dolphins.

The protocol in its final version was submitted to 5 additional experts in rehabilitation / veterinary medicine for review. These reviewers were:

Dr. Katrin Baumgartner (Nuremberg Zoo, Germany)

Dr. Paulien Bunskoek (Dolphinarium Harderwijk, Netherlands)

Dr. Manuel Garcia Hartmann (Marlab, France)

Dr. Kerstin Ternes (Duisburg Zoo)

Dr. Niels van Elk (Van Elk Consultancy, Netherlands)

The authors would like to thank these five reviewers who made excellent comments and suggestions enabling improvement of the protocol.

Additional acknowledgments: Veronica Cendejas, Amanda Ardente, Julieta Olocco, Elaine Reiter, Forrest Townsend, Eric Franks, Rae Stone, Adriana Negron-Olivo, Brenda Bauer, Kristina Martz, Jason Morales, Emanuel Carvalho Ferreira, Carolina Milanesi, Lauro Barcelos (director Museu Oceanografico Prof. Eliézer de C. Rios), the entire technical team of CRAM-FURG, the Federal University of Rio Grande (FURG), the Superintendence of Ports RS, Alejandro Fallabrino, Gabriela Vélez Rubio, Virginia Ferrando, Gloria Mendez de Cabrera (President FMM), Julio Loureiro (Scientific Director FMM), Gaston Delgado and the Technical Staff of the Rescue and Rehabilitation Centre FMM.

This publication was made possible in part by funding from the Organization for the Conservation of Latin American Aquatic Mammals - YAQU PACHA e.V., Nuremberg Zoo, the Association Friends of Nuremberg Zoo (Verein der Tiergartenfreunde Nürnberg e.V.) and NMMF Board of Directors Grants Program, as part of Operation GRACE, Global Rescue of At-Risk Cetaceans and Ecosystems.

Citation: J. Meegan, F. Gomez, A. Barratclough, C. Smith, J. Sweeney, V. Ruoppolo, C. Kolesnikovas, R. Pinho da Silva Filho, P. Lima Canabarro, P. Laporta, J. P. Loureiro, K. Alvarez, S. A. Rodriguez Heredia, A. Cabrera, A. Faiella, A. Saubidet & L. von Fersen – AFCR3, 01-2022

Published by: Alliance for Franciscana Dolphin Conservation Research, Rescue and Rehabilitation (AFCR3)

Cover photo: Marta Cremer, UNIVILLE, Brazil,

Publication layout by: Imre Sebestyén jr / Unit Graphics

Nuremberg, 2022

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Introduction

The Franciscana or La Plata Dolphin (*Pontoporia blainvillei*) is one of the smallest dolphin species and the only river dolphin that inhabit marine waters. It is endemic to the western South Atlantic Ocean, ranging from Espirito Santo State, Brazil in the north to Golfo San Matías, Province Rio Negro, Argentina in the south (Crespo, 2009; Secchi, 2014). Based on genetics and morphometry and for conservation reasons, the range was divided into five management areas (Franciscana Management Areas, Secchi, et al. 2003, Secchi, et al. 2021). Though the causes are unknown the distribution range is not continuous as there are two hiatuses, in other words two "Franciscana -free" areas in the northern range of the distribution.

Due to its preference for coastal waters this species is particularly vulnerable to anthropogenic influences, especially to incidental mortality mainly in gillnet fisheries. Even if exact numbers of Franciscana bycatch for the whole distribution area are still unknown, the high annual mortality estimates that have been calculated for some areas, suggest that bycatch levels are unsustainable (Crespo et al. 2010, Zerbini et al. 2010, Prado et al. 2013). Although other threats exist, potentially compromising the survival of this species, such as habitat degradation, chemical pollution, ingestion of plastic debris and contamination, overfishing and depletion of fish stocks, and climate change, incidental mortality in gillnets is currently the greatest threat to Franciscanas (Secchi, et al. 2021). This dangerous scenario leads to the conclusion that the Franciscana is considered the most threatened small cetacean in South America (Secchi, et al. 2003, Secchi, et al. 2021). As the Franciscana dolphin faces a high risk of extinction it is listed as "Vulnerable" on a global scale by IUCN (Zerbini et al., 2017). Whereas at a regional level, like in Brazil, it is listed as "Critically Endangered" (MMA, 2014).

Due to its status as a threatened species, high unsustainable bycatch rates over the last 40 years and the bleak outlook for its conservation in the future, the Franciscana was selected as one of the focus species in the ESOCC (Ex Situ Options for Cetacean Conservation) Workshop held in Nuremberg, Germany in December 2018 (Taylor, et. al, 2020). The main goal of this meeting was to evaluate to what extent ex situ management can be added to existing conservation strategies in order to secure the survival of species. One major recommendation was to apply a more holistic framework for species conservation planning called the One Plan Approach (Byers, et al., 2013). According to this approach species conservation comprise conservation measures in the natural habitat (in situ) as well as in protected or controlled environments (ex situ). In reality, ex situ approaches comprise a variety of actions including safeguarding animals in protected environments such as semi-natural reserves to prevent species extinction; initiating research programs to fill gaps in our understanding of a species' biology and threats to its survival; rescue and release of stranded individuals; and public engagement programs to promote understanding and support of species conservation.

It is generally agreed that especially in the case of the Franciscana there are still many information gaps that need to be filled in and many open questions that need to be answered. By incorporating methodologies and tools developed and used in ex situ circumstances, many of these unknowns could be clarified.

Dolphins stranded alive deserve special attention. These phenomena are observed worldwide and subjectively the impression is that the number of live strandings is increasing. In the case of Franciscana dolphins, it is primarily neonate calves that strand alive. The following graph (Fig. 1) illustrates the extent of the problem; since 1991, a total of 133 Franciscana dolphins stranded alive have been recorded. Of these, only four were adults, one was a juvenile and the rest were neonates (128 animals, data from Fundación Mundo Marino, Argentina; CRAM-FURG, AIUKA and 3R from Brazil, 2021). However, these numbers do not refer to the entire range of the species, but only to certain limited coastal areas in Argentina, Uruguay and Brazil. It can be assumed that the number of stranded animals may well be higher when considering the entire range. However, such a calculation is not possible due to lack of data.

A protocol published in 2022 was mainly dedicated to calves stranded alive. In this current protocol, we focus on juveniles and adults, which, as mentioned above, are not as common but still require special care. To date, rehabilitation success of stranded Franciscana dolphins is minimal and there is a critical need for enhancement of current conventions. The main goal of this protocol is the development of a well-planned and scientifically based rehabilitation routine to increase individual animal survival. When an endangered species is in need of help, returning just one individual animal to a population can make a difference.

Rehabilitation also offers the opportunity to obtain previously unknown aspects of the biology through research and proper data collection. It is therefore equally important to outline through this protocol the framework for good scientific work. When rehabilitation is combined with research the conditions are in place to improve our understanding on the ecology, behavior, management, handling and health of this species. Finally, it is also important to note that animals who are undergoing a rehabilitation process can be used as drivers for education and outreach programs, highlighting conservation needs. A live animal in rehabilitation due to environmental issues or other threats can serve to draw attention to conservation problems for their species as a whole. In summary, we hope that these protocols will primarily serve to increase the likelihood of successful rehabilitation of threatened Franciscanas, providing a good basis for knowledge enhancement and education.

An important aspect to mention is that these protocols were developed with the

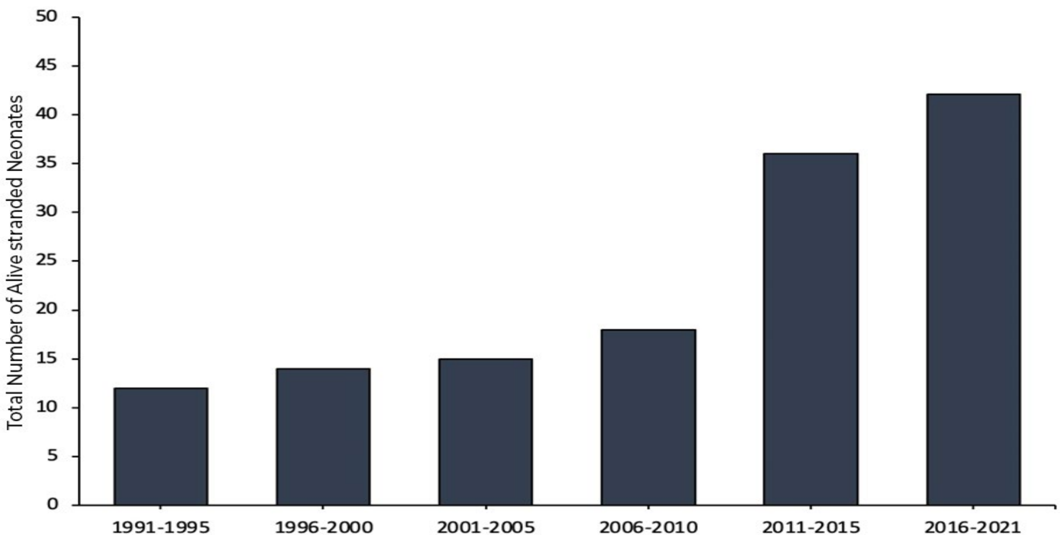

Figure 1: Number of live stranded Franciscana dolphins found in some areas of Argentina, Uruguay and Brazil since 1991. (Source: Fundación Mundo Marino, Argentina; CRAM-FURG, AIUKA and 3R, Brazil)

participation of numerous specialists in the field of marine mammal science, medicine, rehabilitation and behavior. The South American colleagues have worked in centers in their home countries and have been involved with the rehabilitation of Franciscana for many years. Protocol development followed a gradual, collaborative process pulling on local and species expertise, as well as expertise developed working with other small cetaceans. The neonate protocol was partially applied during the stranding season of 2020, 2021 and 2022, when seven Franciscana dolphins were found stranded in Argentina and Brazil. This practical application during protocol development helped not only to test the protocol for its applicability, but also to refine it. Protocol development also included the standardization of live and dead animal data collection, allowing for data gaps to be filled through each stranded animal. Moving forward these protocols will serve as "living documents" to be updated regularly as lessons are learned through each rehabilitation experience.

Future considerations for the successful reintroduction of rehabilitated Franciscana dolphins

Recovering and rehabilitating small cetaceans from a stranding event had limited success historically. To the best of our current knowledge, this protocol describes an attempt to triage and rehabilitate Franciscana dolphins during various stages of rehabilitation. If the rehabilitation efforts are successful, it is expected that individuals will be medically healthy to potentially be returned to the ocean. Determining fitness for release back to the sea would require a separate decision-making process and guidance from additional resources such as the Guidelines for the Safe and Humane Handling and Release of Bycaught Small Cetaceans from Fishing Gear - https://www.cms.int/en/publication/guidelines-safe-and-humane-handling-and-release-bycaught-small-cetaceans-fishing-gear

For many other species where individuals have received rehabilitation under similar circumstances, reintroduction to the wild, is the ultimate goal. In the rehabilitation of aquatic mammals with subsequent release, there are different experiences in terms of success with successful and difficult cases. Adimey et al. (2016) found a 72% success rate in the release of rehabilitated manatees, particularly wild-born manatees. Comparable, though with less success (46.3%), was observed in rehabilitated sea lions (Gage et al., 1993). Rehabilitation of stranded cetaceans is particularly difficult. As was shown in the study by Zagzebski et al. (2002), where only a minority survived after rescue.

Moore et al. (2007) point to the divided response to marine mammal rehabilitation. Challenges include conflicts with fisheries, lack of ecological understanding of recipient populations and potential harm to human health. Nonetheless, rehabilitation provides an opportunity for institutions to demonstrate their commitment to animal welfare, advance research and raise awareness of animal conservation. In some cases, such as the Franciscana dolphin, rehabilitation is a priority because of the threatened populations. In summary, the success of odontocetes rehabilitation still has room for improvement, highlighting the need for robust protocols and assessments.

It is particularly important to document the success through a follow-up, only tracking the animals by means of transmitters can show whether reintroduction has been successful. Especially in the context of the One Plan Approach and in line with the International Union for Conservation of Nature's (IUCN) Integrated Conservation Planning for Cetaceans (ICPC) group, who advocate an integrated conservation approach for cetacean species, it is essential to leave all options open regarding the future of successfully rehabilitated Franciscana dolphins.

Rehabilitated Franciscanas that have spent a long time in the care of humans are certainly a great challenge because these animals rarely have contact with conspecifics and may become habituated to humans. Despite these difficulties, the final goal for these rehabilitated animals should be a life in a natural or semi-natural habitat. Like those used in China for the government's Yangtze finless porpoise (*Neophocoena asiaeorientalis asiaeorientalis*) conservation program, semi-natural ex situ reserves offer one possibility. Investigations are continuing into local areas in-country that are in close proximity to the Franciscana habitat where wild conspecifics are present. Opportunities for long-term care of rehabilitated Franciscanas in semi-natural reserves may emerge with the ultimate goal of reintroduction to the wild. These areas offer potential habitat where the rehabilitated animal can acclimate to the natural soundscape, hunt prey fish, and experience the climatic conditions and challenges of the wild. This type of gradual acclimatization and soft-release measures have been used for many species and it is conceivable that rehabilitated Franciscana dolphins may be excellent candidates for such a program.

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RESCUE AND REHABILITATION FOR JUVENILE/ADULT FRANCISCANA DOLPHINS

(*Pontoporia blainvillei*)

In the event that a juvenile/adult Franciscana dolphin is stranded, the following protocol may be applied

INITIAL ANIMAL ASSESSMENT AT STRANDING LOCATION

- The time between the stranding and the arrival of the rescue team can be critical to relieve distress and improve the chance of survival.

- Educate the local public (personnel on-site) who may be waiting with the animal before the rescue team arrives of some basic tips to care for the dolphin:
  - Ensure human safety and awareness around animal
  - Do NOT attempt to release the animal back in the ocean
  - Do NOT lift the animal out of the water unless absolutely necessary
  - Keep the animal upright with the blow hole above the water at all times
  - Minimize handling of the animal
  - Reduce the noise and physical contact with animal to reduce stress
  - Protect eyes from debris (e.g. sand)
  - Animals on the beach must be protected from harsh weather:
    - Cover skin with wet towels to prevent skin damage and exposure dermatitis
    - Hyperthermia can occur with prolonged exposure to warm temperatures and hypothermia can occur with cold temperatures
    - Provide shade from the sun to protect the skin from solar dermatitis
  - Avoid maneuvering pectoral fins to prevent bone fractures or joint damage
  - Dig sand underneath flippers to reduce respiratory distress or place in a shallow pool of water (Figure 2) while waiting for the rescue team

- It also important to educate the public or any personnel on-site about the importance of zoonotic diseases in order to ensure that the humans are protected because this is a sick animal that could potentially spread illness and is provided with protective gear such as gloves, facemasks, etc

- When the rescue team arrives, the veterinarian/rescue team lead will make an initial assessment and will have medical supplies to provide supportive care

Figure 2. Example of a helpful option to provide the calf a shallow area of water while waiting for the rescue team to arrive (Photo credit Fundación Mundo Marino)

- The most appropriate location to perform the initial assessment and supportive care treatments should be determined by the rescue team on site

- Ideally perform the initial assessment *prior* to moving or transporting the animal, in order to establish a *baseline* before moving the animal, however it may be more practical to first remove the animal from a crowded or unsafe location and perform the initial assessment during the transport

- Initial assessment at the stranding location should be brief:
  - Note vitals: respiratory rate, heart rate, temperature
  - Assess behavioral responses:
    - Alert: aware, responsive to stimuli
    - Weak: responsive only after intense stimuli
    - Non-responsive: not responding to touch or noise
  - Examine for external injuries that may need immediate attention
    - Measure body length to determine age class (Appendix 1). A standard straight length is defined as the measurement of the straight-line distance from the tip of the rostrum to the notch of the flukes

POSSIBLE STRANDING SCENARIOS

Assess the situation to determine if the dolphin is a candidate for rehabilitation or release:

Scenario #1: Animal appears injured or ill and requires rehabilitation (NOT a candidate for release).

- ensure the animal is stable (may need immediate supportive care), and transport to rehabilitation facility as soon as possible

Scenario #2: Animal appears healthy (alert, normal vitals, no obvious external injuries) and IS a candidate for release

- Consider immediate release if healthy or after receiving supportive care
- If available, apply a tracking tag and release from site
- If other healthy dolphins are also present, release them together

Scenario #3: Animal is severely injured warranting euthanasia (if permitted in your county)

- Prior to euthanasia consider the following:
  - If lactating female consider collecting milk/colostrum from the dam
  - Collect blood (IgG/serum/plasma) for analysis and archiving

- Transfer to rehabilitation facility for full necropsy examination and sample collection

Scenario #4: Animal is recently deceased
Transfer to rehabilitation facility for full necropsy examination and sample collection

TRANSPORTING THE DOLPHIN BACK TO THE RESCUE FACILITY

- It is important to transport the animal to the rehabilitation facility as quickly as possible in order to provide medical care and diagnostics

- Determine if the dolphin is stable prior to transport:
  - If the animal appears extremely weak or non-responsive it may need immediate supportive care prior to or during the transport, especially if the duration of the trip is long (> 60 minutes)
  - Stranded juvenile/adult dolphins can be susceptible to *dehydration*, so it may also be necessary to administer fluids *prior* to or *during* transport. Please refer to the "**Treatments and Medications**" section later for details on intervention
    - Avoid oral hydration prior to transport as this may lead to vomiting and aspiration

- If the decision is made to bring the dolphin back to the rescue facility for medical care, the stranding response team/crew will have means to transport the dolphin

- The preferred method for transport is in water in a transport container (Figure 3.):
  - The transport container should have either a fleece-lined sling and soft padded foam on the bottom
  - Fill the transport box with a small amount of water to keep the eyes submerged below the water level and float the animal to aid in buoyancy. Keep the water level well below the blowhole due to risk of aspiration during transport

Figure 3: Transport container designed for a small cetacean (Photo credit NMMF/VaquitaCPR, AIUKÁ)

- If a transport container is not available, the animal can be placed on wet soft foam padding or an air mattress
- Wet the animal at all times during transport, being careful to avoid water in the blowhole
- Monitor the animal very closely at all times during transport
- Please refer to **Appendix 2. Animal Monitoring Sheet During Rescue and Transport** to record the data
  - Record respiratory rate and breath quality during transport (# of breaths per 5 minutes)
    - Normal respiratory rate: 5-25 breaths per 5 minutes
    - Abnormal respiratory rate: < 5 breaths per 5 minutes *OR* > 25 breaths per 5 minutes
    - Ideally record the respiratory rate *continuously* during the transport
    - How often to record will depend on the duration of the transport and if the respiratory rate is normal or abnormal
  - Record heart rates during the transport (# of beats per minute)
    - Normal heart rate: 80-140 beats per minute (bpm)
    - Abnormal heart rate range: < 70 bpm and > 150 bpm
    - The heart rate can be measured by gently palpating the heart beating near the ventral surface of the chest near the axillary region or by auscultation using a stethoscope
    - If measuring the heart rate is causing too much stress to the animal during the transport then please obtain a heart rate when safely possible and continue to closely monitor the respiratory rate
  - Record behavior and activity
    - Monitor closely for signs of extreme stress or shock such as: arching, foam upon expiration, extreme restlessness with tachycardia and tachypnea or extreme passiveness with loss response to environmental stimuli, flaccid jaw tone and bradycardia may indicate intervention is warranted.
  - Monitor ambient air temperature during the transport to prevent hypothermia or hyperthermia
    - Adjust the ambient temperature during transport or provide warm or cold wet towels if the temperature too warm: > 80F (> 27 Celsius); or too cold < 60 F (< 15 Celsius)

- If the animal is being transported in water, record water temperatures and maintain between (20-25C)
  - Salt water is preferred but fresh water may be used for short periods of exposure (<12 hrs)

- If the dolphin is transporting with multiple animals, keep them in close proximity whenever possible in an attempt to keep animals communicating and more comfortable.

- Sedation during transport may be administered at the discretion of the attending veterinarian
  - If efforts to reduce stress and anxiety have been unsuccessful, and if everything else has been corrected and the dolphin is still displaying signs of distress, sedation may be warranted
  - Diazepam (0.05-0.1 mg/kg) or midazolam (0.04 - 0.08 mg/kg) can be administered at IV or IM to reduce anxiety and provide muscle relaxation, unless the cetacean is already very depressed or is a very young calf in which case this would be contraindicated
  - If sedation with diazepam/ midazolam is used, be sure to also have the reversal drug (flumazenil) on hand should complications arise and the sedation needs to be reversed

ARRIVAL TO THE RESCUE/ REHABILITATION FACILITY:

- Upon arrival to the rescue facility, place the dolphin in a round/oval pool to facilitate close monitoring and effective handling as needed for assisted feeding and animal care

- If the dolphin is too weak or unable to swim in a normal upright position, a rescue staff member may need to continuously guide the dolphin around the pool, supporting the animal's weight, and keeping the blowhole above the water surface (Figure 4.)

- It is very important to attempt to keep the eyes under the water as much as possible

- Clockwise and anti-clockwise movement should be encouraged to reduce the risk of muscle damage, contractions, spasms, or spinal curvature

- Custom flotation devices/stretcher can be used to aid in buoyancy (Figure 5.)
  - It is important to monitor closely for pressure wounds on the skin that may be caused by the harness/ flotation device

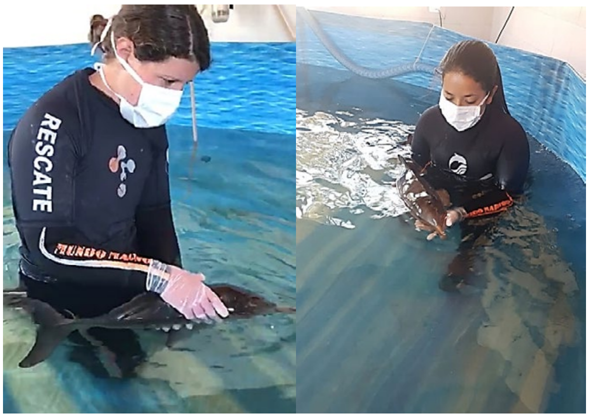


Figure 4. Examples of rescue staff members guiding the dolphin around the pool, gently supporting the animal's weight, with the eyes below the water and the blowhole above the water surface (Photo credit Fundación Mundo Marino)

  - The flotation device should allow the dolphin to lower its head and eyes under the water while keeping its blowhole above the water, the body should appear relaxed and as low in the water column as possible

  - If a flotation device is used, please only use for short periods of time during the day if possible, and remove the dolphin from the flotation device regularly to prevent skin damage, muscular curvature and scoliosis

- Keep exposed areas of skin wet at all times, apply ointments such as lanolin or Vaseline (petroleum jelly) to the skin to keep the skin from drying/cracking

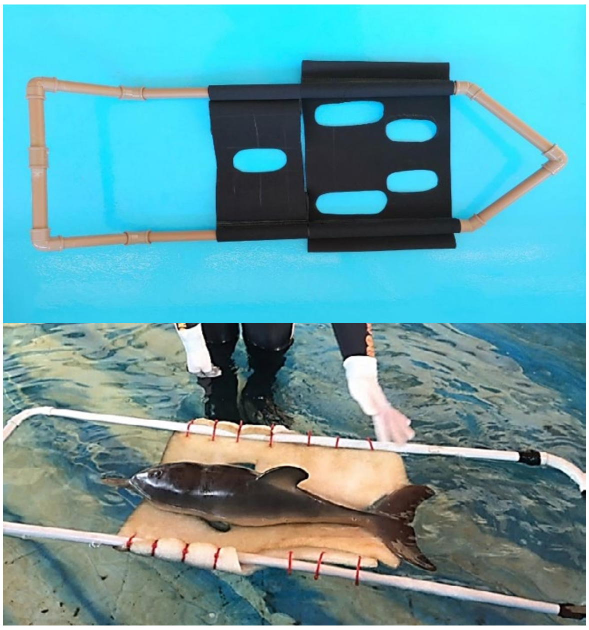


Figure 5. Franciscana calf shown in a custom stretcher made of soft fabric with holes for the pectoral fins and the tail, and eyes are effectively under the water. It is important to keep the exposed skin wet/moist at all times. [Photo credit Aiuká / CRAM-FURG (left); Fundación Mundo Marino (right)]

IMPORTANT CONSIDERATIONS FOR HANDLING THE DOLPHIN:

- Avoid excitement, loud noises, and struggling when handling the dolphin

- IMPORTANT: Avoid removing the dolphin from the water unless absolutely necessary
  - ALL feedings, treatments, medi-cations, and blood collections should ideally be performed in the water
  - Any out of water procedure should be as brief as possible
  - If you need to remove the dolphin from the water, continuously monitor vital signs (HR and RR) and behavioral activity and return animal immediately to the water if these become abnormal
  - Take the time to plan any out of water procedure with the team in advance to create familiarity with it

- Signs of stress during handling include tachycardia (persistently >120bpm) or bradycardia (persistently <60 bpm), arching of the body, extreme struggling, fluke slapping, irregular, shallow, or rapid breathing, holding blow hole open, opening mouth and regurgitation, sudden weakness, cessation of vocalizations

- At any time during an intervention procedure with a dolphin, personnel must be willing and ready to abort the procedure if concerns for the status of the dolphin are observed or due to significant changes in vital signs.

- The handling must be stopped if these signs of distress are observed during any procedure

- It is recommended that, especially in the first weeks, only experienced staff in cetacean handling and rehabilitation care for the animal

- Furthermore, it is important that the same staff takes care of the animal during the first phase of rehabilitation, if possible. Experience has shown that a constant change in the care team causes stress for the animal.

PHYSICAL EXAMINATION: (*AFTER ARRIVING TO RESCUE FACILITY*):

- The next step will be to administer supportive care and perform an initial medical examination
- The animal will likely need immediate supportive care to include oral fluids, medications, etc. Please refer to the "**Treatments and Medications**" sections below for more details
- A medical examination should be performed as soon as possible after the animal arrives at the facility so that therapy can begin immediately
- The initial examination on Day #1 may need to be brief, and should at *minimum* include vitals, blood sample, weight, and a visual examination
- Please complete the physical exam form as provided (**Appendix 3. Physical Examination Form**)

- **Morphometrics**
  - Collect a body weight
    - Removing the dolphin from the water to collect a body weight should be very brief.

Helpful tips for collecting the weight: place the scale immediately adjacent to the pool. A person stands/sits on the scale and people in the water gently lift the dolphin and transfer the dolphin to the person standing on the scale. This process should take only 1 minute.

- The dolphin can also be gently placed onto a small food/animal scale to collect the weight
- Girth - axial girth taken behind the pectoral flippers.
- Length: Measure body length to determine age class. A standard straight length is defined as the measurement of the straight-line distance from the tip of the rostrum to the notch of the flukes.

See additional **Appendix 1: Growth Curve-Age estimation for Franciscana dolphins**

Figure 6. Collecting a body weight: A person stands/sits on the scale and gently holds the calf (left) or place the calf on a small scale (right). Removing the dolphin from the water to collect a body weight should be very brief. [Photo credit Fundación Mundo Marino (left); AIUKÁ / CRAM-FURG (right)]

- Vitals
  - Respiration rate: record # of breaths/5 mins
    - Normal respiratory rate: 5-25 breaths per 5 minutes
    - Abnormal respiratory rate: < 5 breaths per 5 minutes *OR* > 25 breaths per 5 minutes
    - Reported Franciscana respiratory rate: 5-9 per minute (Baldassin, et al. 2007)
    - Please record once for the physical exam form (**Appendix 3**)
    - For further details on daily frequency refer to the daily monitoring section
  - Heart rate:
    - Normal heart rate: 80-140 beats per minute (bpm)
    - Abnormal heart rate range: <70 bpm and >150 bpm
    - Reported Franciscana heart rate has been reported to range from 99 to 139 beats per minute (Baldassin, et al. 2007)
    - Please record once for the physical exam form (**Appendix 3**)
    - For further details on daily frequency refer to the daily monitoring section
  - Temperature:
    - Collect a rectal body temperature for the physical examination form (**Appendix 3**)
    - Only temperature reported for Franciscana was 36.4 C (97.5 F) (Baldassin, et al. 2007)
    - Normal temperature range for Tursiops truncatus: 36-38 C (96-100F)

- **Blood Sampling Collection Method**
  - Blood can be collected from the periarterial venous rete (PAVR) from either the fluke blade or from the caudal peduncle using a 21-25 gauge butterfly and a 3-cc syringe

- It is strongly recommended that the dolphin remain in the water for the blood collection. This can be accomplished by one handler supporting the body and head, and a second handler gently lifting the tail out of the water for the third person who is collecting the blood sample. Blood can be collected more easily on the ventral aspect of the tail fluke but dorsal sampling is also an option if positioning is challenging.

- During the blood collection monitor the respiratory rate and heart rate very closely, and abort the procedure if the dolphin shows any signs of distress or if the parameters become abnormal

- Clean the blood collection site (using dilute betadine or chlorhexidine and alcohol), then immediately cover the site with antibiotic ointment and a gauze after removing the needle until a clot (hemostasis) is achieved

- **Blood tests to analyze:**
  - It is critical that the turn-around time for blood results be as fast as possible
  - Ideally all of these tests should be performed on-site at the rehabilitation facility. If this is not possible, then ideally identify a laboratory that can ensure rapid test results any day of the week.
  - **Tests to perform *on-site at rescue facility for immediate results***:
    - Blood glucose using a hand-held glucometer (normal > 60)
    - Blood smear to perform a differential
    - Hematocrit and total protein
    - Estimated white blood cell count
    - Erythrocyte sedimentation rate (if available)
    - Blood gas: if available
  - **Tests to send to the laboratory:**
    - Complete blood count (CBC): WBC, Hct, RBC, Hct, Hgb, etc

- Chemistry panel: glucose, electrolytes, BUN/Creatinine, minerals, iron, albumin, globulin, alkaline phosphatase, transaminases, bilirubin, fibrinogen, etc

- Archive (freeze) serum if possible
- Additional blood tubes may include but not limited to: DNA, Cortisol, aldosterone, etc

Figure. 7. Blood collection from the superficial vessels of the ventral fluke blade (top) and from the central caudal peduncle (bottom) (Photo credit Fundación Mundo Marino)

Table 1. Proposed Blood Collection schedule

Day 1: Collect the first blood sample to establish baseline data and to help determine the most appropriate medications and treatments
Week 1: During this first week, be prepared to collect blood daily if needed*** At a minimum, collect blood on: Day 1, Day 3, Day 7
Week 2: collect a minimum of one follow-up blood sample, more may be indicated
Week 3: collect a minimum of one follow-up blood sample, more may be indicated
Week 4: collect a minimum of one follow-up blood sample, more may be indicated
Month 2+: collect a follow-up blood sample every 2 weeks, if the animals is doing well. Be prepared to collect blood more often if needed***
If the outside laboratories are closed for any reason, at a minimum, be prepared to collect blood and perform the on-site blood tests described above if the animal is sick or a blood sample is warranted

- Full body ultrasound:
  - If available perform a thorough ultrasound examination of the thoracic cavity:
    - Examination of the lungs should be performed to evaluate for pneumonia, pulmonary edema, pleural effusion, or other abnormalities
    - Evaluate the heart to monitor for pericardial effusion, contractility, etc
  - Perform a thorough ultrasound examination of the abdominal cavity
    - Examination of the gastrointestinal tract should be performed to evaluate gut motility, obstructive patterns, intestinal gas, gastroenteritis, ascites, etc
    - Thorough examination of the reproductive tract if female to rule out pregnancy

- Perform hearing/acoustic testing if the equipment is available (Auditory Evoked Potential-AEP) to determine if the animal has any hearing deficits to determine if they are a candidate for release

- Obtain photographs (minimum L/R of dorsal fin and other distinguishing features)

- Additional biological samples should be collected for diagnostics:
  - Collection of respiratory sputum/ blowhole samples should be submitted for bacterial and fungal culture at a minimum
  - Feces and gastric samples should be submitted for gross and microscopic cytologic evaluation as well as bacterial and fungal cultures
  - Other samples for collection may include: any discharge/abscess fluid, skin lesions, urine, etc
  - Archive frozen samples for potential additional testing if warranted

Disclaimer: Sampling decisions will be made by the veterinarian based on dolphin stability throughout the exam. With the exception of weighing, all other procedures will be undertaken while the animal is supported in the water. The examination will be aborted if the animal is deemed unstable at any point. Any additional sampling will be done at later physical exams once animal acclimation and stability has improved.

FEEDING AND NUTRITION

Refer to Table 2. Proposed feeding schedule

- During the first day (Day 1):
  - When the dolphin first arrives, it is important to first correct dehydration
  - Initially hydrate the animal with oral electrolyte
  - Calculating stomach volume: 2 ml/kg
  - Administer oral fluids every 2-4 hours
    - The ideal frequency will depend on the age of the animal, the hydration status, and the health of the gastrointestinal tract
  - Initially start with small volumes (~ 20-30 mL) to avoid risk of aspiration
    - Slowly increase the volume as the animal grows and becomes more acclimated to tube feeding; being careful to avoid vomiting or aspiration
  - Determine the best estimate of the age of the animal to help determine the most appropriate diet and feeding frequency
  - In the wild, Franciscana dolphins are weaned at approximately 8 months to 1 year of age, therefore juvenile and adult Franciscana dolphins will be primarily on an all-fish diet in the wild
  - The best source of nutrition for juvenile/adult dolphins is whole fish
    - Please offer whole fish (live and dead) several times per day to encourage the dolphin to begin eating on its own
    - Refer to **Appendix 8. Stomach contents free-ranging Franciscana dolphins – based on length and age** for more details
    - Franciscana dolphins feed on a wide variety of mainly bottom dwelling fish species. Where possible attempt to feed fish species naturally consumed in the wild
  - Tips on how to re-introduce fish during rehabilitation
    - Offer live fish (partially disable/ injure the fish to slow them down)
    - Assist feeding of frozen dead fish
    - "fish school" with frozen fish: fish are placed on tongs or tossed into the pool
    - It is important that the animal does not become too acclimated to humans during the feeding process
  - If the dolphin won't eat whole fish initially, supplement the diet daily with a blended fish gruel
  - The dolphin can be tube fed whole fish blended with electrolyte solution to provide nutritional support however a balance must be achieved between the stress of handling and the benefit of nutrition
  - Calculating the caloric requirements:
    - Kcal goal: 100-150 kcal/kg/day
  - Gruel recipes:
    - Refer to **Appendix 4. Recipe for fish gruel** for detailed instructions on preparing the fish gruel
    - Prepare the gruel daily, and store in the refrigerator until use
    - Discard any unused gruel after 24 hours
    - Initially provide gruel in lower concentrations, diluting with electrolytes or water
    - Steadily increase the concentration of the formula/water ratio
  - Gruel feeding delivery methods:
    - Gruel or other liquid solutions can be administered via orogastric tube attached to a feeding tube
    - Use a small flexible rubber/ silicone stomach tube with a smooth rounded tip
      - Size of the tube will initially be small, but consider increasing tube size as animal grows and

becomes more acclimated to tube feedings
  - Preferred tube size: ~ 6 mm diameter (18 French); Length: 16 inches (41 cm)
  - Select a tube material that is flexible, but rigid enough to pass easily into the stomach without kinking or bending
- Determine appropriate tube length by measuring the distance from the end of the rostrum to the anterior insertion of the dorsal fin and marking the tube with tape or a permanent marker
- Insert the lubricated tube into the stomach (using the marked length) (Figure 8.)
- Administer the gruel/liquids very slowly to avoid vomiting or aspiration

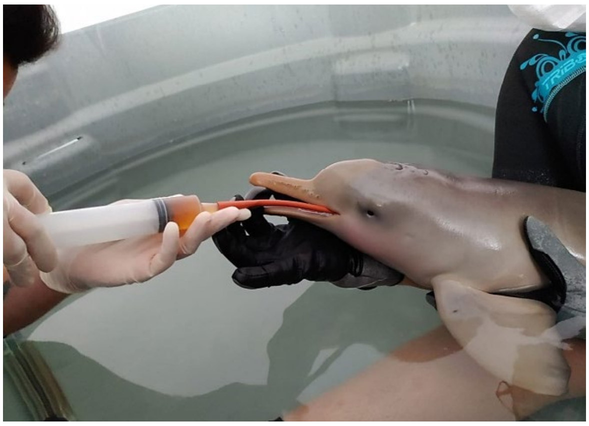


Figure 8. Tube feeding a neonate Franciscana. (Photo credit CRAM-FURG)

Table 2. Proposed Feeding schedule

Date	Diet	Record amount eaten	Feed frequency
Day 1	Initially hydrate with electrolyte water (oral/ subcutaneous) +/- glucose	Kg of fish: Volume of gruel:	Hydrate every 2-4 hours
Day 2	Offer whole fish (frozen/live) *If animal won't eat fish, supplement with gruel: begin 25% fish gruel + electrolyte water	Kg of fish: Volume of gruel:	Feedings every 2-4 hours
Day 3	Offer whole fish (frozen/live) *If animal won't eat fish, supplement with gruel: increase to 50% fish gruel +electrolyte water	Kg of fish: Volume of gruel:	Feedings every 2-4 hours
Day 4	Offer whole fish (frozen/live) *If animal won't eat fish, supplement with gruel: increase fish gruel to 75% concentration	Kg of fish: Volume of gruel:	Feedings every 2-4 hours
Day 5-7	Offer whole fish (frozen/live) *If animal won't eat fish, supplement with gruel: increase fish gruel to 100% concentration	Kg of fish: Volume of gruel:	Feedings every 2-4 hours
Week 2	Offer whole fish (frozen/live) *If animal won't eat fish, supplement with fish gruel	Kg of fish: Volume of gruel:	Feedings every 2-4 hours
Week 3	Offer whole fish (frozen/live) *If animal won't eat fish, supplement with fish gruel	Kg of fish: Volume of gruel:	Feedings every 2-4 hours
Week 4	Offer whole fish (frozen/live) *If animal won't eat fish, supplement with fish gruel	Kg of fish: Volume of gruel:	Feedings every 2-4 hours
Month 2+	Offer whole fish (frozen/live) *If animal won't eat fish, supplement with fish gruel	Kg of fish: Volume of gruel:	Feedings every 2-4 hours

TREATMENTS AND MEDICATIONS

Hydration therapy:

- Stranded dolphins can also be susceptible to dehydration, so it may also be necessary to administer fluids in addition to adding fluids to fish to maintain adequate hydration status
- Fluid type: use a balanced electrolyte crystalloid solution such as Lactated ringer's (LRS), 0.9% saline
- Fluids can be administered either subcutaneously, orally, or intravenously
  - Any combination of these routes is also acceptable
- Total daily hydration requirement is approximately 40-50 ml/kg/day
- If the daily hydration requirement cannot be achieved through the oral feedings, supplemental hydration can be administered at 10 ml/kg IV/SQ once per day to prevent dehydration

Glucose therapy:

- Juveniles/subadults with anorexia or emaciation may be susceptible to hypoglycemia, so it is important to monitor blood glucose closely during the initial exam until eating and appetite is fully established
  - Blood glucose can be easily collected with just a few drops of drop of blood and using an over-the-counter handheld rapid blood glucose meter
  - Hypoglycemia is present if the blood glucose is < 60 mg/dl
  - ***If the glucose is < 60, administer a glucose solution described here, then repeat the test. Continuing administering glucose until the glucose level normalizes
- Oral route:
  - Administer a 5% glucose solution orally
  - Dose: 2 ml/kg
  - The concentration of glucose is dependent on the blood glucose level
- Subcutaneous route:
  - Use a 2.5% glucose solution
  - Dose: 10 ml/kg
- Intravenous route:
  - Use a 2.5-5% glucose solution
  - Dose: 10 ml/kg
- How to make dextrose solutions:
  - To make a 2.5% solution: add 50mL of 50% dextrose (or 25g dextrose) to a 1L bag of fluids (NaCl or LRS)
  - To make a 5.0% solution: add 100mL of 50% dextrose (or 50g dextrose) to a 1L bag of fluids (NaCl or LRS)

Injection techniques:

- Clean the IM/SQ injection site (using dilute betadine or chlorhexidine and alcohol), then immediately cover the site with antibiotic ointment and a gauze after removing the needle until a clotting (hemostasis) is achieved and to prevent the medication from leaking out of the site
  - Intramuscular and subcutaneous injections can be given at the following sites (Figure 9.).

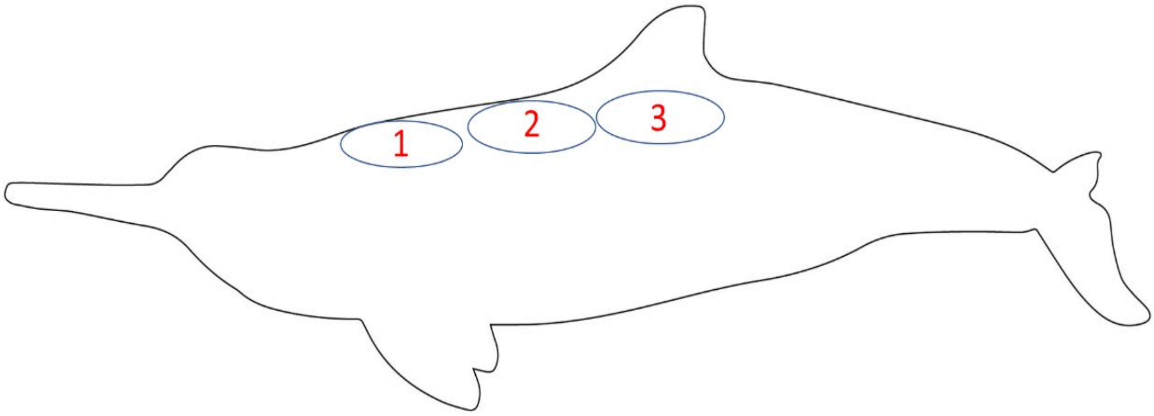
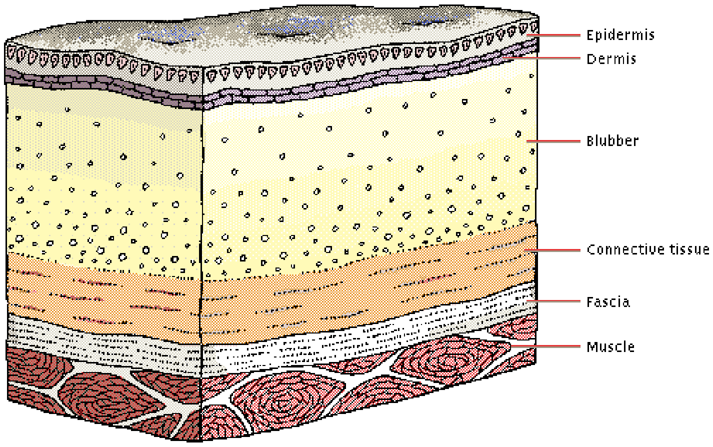


Figure 9. Injection sites for the Franciscana dolphin
Site 1- cranial subcutaneous space and cranial epaxial muscle***
Site 2- middle epaxial muscle and subcutaneous space***
Site 3- caudal epaxial muscle
***IMPORTANT: ideally use ultrasound to first measure the depth of the subcutaneous space and the muscle layer to determine the most appropriate needle length to avoid injuring deeper organs (lung)

Subcutaneous space blubber depths for Franciscana dolphins range from:
Juvenile: 1.2-2.5 cm (0.5- 1 inch)
Adult: 1.5-3.0 cm (0.6 – 1.1 inch)

Muscle layer depth for Franciscana dolphins range from:
Juvenile: 2-4 cm (0.8 – 1 inch)
Adult: 2.4-4.8 cm (1 – 1.75 inch)



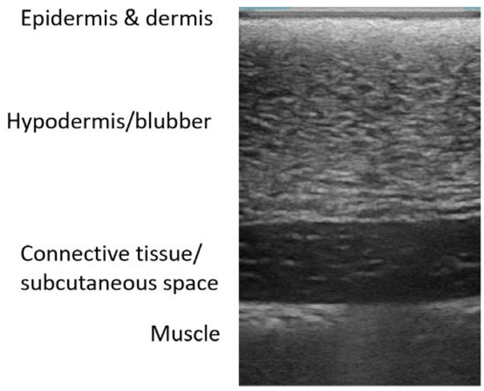


Figure 10. Schematic and ultrasound cross sectional images of a small cetacean depicting the skin, subcutaneous, and muscle layers.

MEDICATIONS

- Please refer to **Appendix 5. Drug Formulary** for a detailed list of drugs, doses, and routes
- Upon arrival to the rescue facility, administration of a **broad-spectrum antibiotic** is recommended
- Additional medications may also be warranted:
  - Gastrointestinal medications such as anti-gas, antacid or anti-nausea medications may also be required
  - Antifungal medications may be warranted if a primary fungal infection is suspected or for prophylaxis for long-term antibiotic use
  - Furosemide (diuretic)

    - Potential indicators: Froth/fluid exiting the blowhole; tachypnea, pulmonary edema seen on ultrasound/xray
    - Use with caution if the patient is dehydrated and avoid multiple doses as diuretics can lead to severe dehydration and electrolyte derangements
  - Steroids:
    - Low-dose, single dose administration may be warranted for inflammation, post-stranding distress, other
    - Use with caution and avoid multiple doses if possible as immunosuppression can occur with overuse

MEDICAL RECORD KEEPING

Please refer to Appendix 6. Example of a daily monitoring data sheet

Once animals are under human care, animal care staff will observe and monitor their behavior 24 hours/day. Record keeping will be implemented immediately in all areas of animal care, to include but not limited to, behavioral and physiological response, vital signs, feeding, medical care, and social behavior.

Daily monitoring (Record the following data every day):

- **Respiration rate:**
  - Record # of breaths/5 minutes
  - How often to record:
    - Day 1: record respirations hourly
    - Week 1: record respirations hourly
    - Week 2-4: record respirations a minimum of 3 times per day if normal
    - Month 2+: record respirations a minimum of twice per day if normal
- **Heart rate:**
  - Record the number of beats per minute
  - How often to record heart rate:
    - Day 1: record heart rate a minimum of three times per day
    - Week 1: record heart rate a minimum of once per day if normal, more often if abnormal
    - Week 2-4: record heart rate a minimum of once per day if normal
    - Month 2+: if the animal is still being handled for feedings record heart rate once per day
- **Behavioral monitoring:**
  - Describe behaviors: swim patterns, vomiting, crunching, arching, independence (**Appendix 10**).
- **Defecation:**
  - Note the frequency of defecation, color, and consistency are important to monitor. Feces should be a thick liquid, light green in color, and should not float or exhibit gas bubbles.
- **Body weight:**
  - Day 1: obtain an initial body weight when the animal arrives to the rehabilitation facility
  - Week 1: obtain a weight twice per week, more often if the animal is not gaining weight
  - Week 2-4: obtain a weight once per week, more often if the animal is not gaining weight
  - Month 2+: obtain a weight every other week, more often if animal is not gaining weight

WATER QUALITY

- Please refer to **Appendix 7. Water Quality Monitoring Data Sheet** to record data

- When the dolphin first arrives to the rescue facility, place the animal in a round (15 feet diameter) or oval shaped pool (15 feet x 10 feet) with a preferred volume of 7,000-10,000 Liters

- Maintain salinity in pool at 20 ppt or higher
  - 20 ppt = 20 grams salt per Liter

- Maintain water temperature at 25-28 Celsius (77-82 degrees Fahrenheit) to prevent shivering and to promote skin healing

- Please monitor the following water quality parameters <u>**every day**</u> and record on the form
  - Water temp: 25-28 Celsius
  - Salinity: 20-35 ppt
  - pH: 7.2-8.4
  - Ammonia < 2 ppm
  - If chemical filtration is used, these chemicals will need to measured daily as well (i.e. chlorine, ozone, etc)

- Please monitor the bacterial coliform count <u>**once per week**</u> and record on the form
  - coliform count must be < 1,000 MPN (most probable number)
  - **if the coliform count is > 1,000 = increased fecal bacteria in the water, this requires a pool cleaning and a water change of at least 25% then repeat the test

- We recommend 25% water changes every other day or as needed to keep the water clean and sanitary. **Please also siphon any debris/sediment on the bottom of the pool and the pool walls may also need to occasionally be cleaned with a scrub brush

- Mechanical filtration:
  - Utilizing a mechanical filtration system is highly recommended, as it will help to remove any debris or solid waste from the water
  - Examples of mechanical filtration may include a cartridge filter, sand filter, etc
  - Please clean the filtration system regularly (the frequency will depend on the type of filter used and the water quality parameters)

- Chemical filtration (i.e. chlorine/ozone)
  - Please use chemical filtration with CAUTION and ONLY if your facility has previous experience with these chemicals
  - Do NOT use unless you have the ability to measure chlorine or ozone levels daily as exposure to elevated levels can be harmful/toxic
  - If using chlorine: a low range of 0.2-0.5 ppm could help to maintain water sanitation to reduce the frequency of water changes

CRITERIA FOR RELEASE

- It is important to first determine that the animal is a healthy candidate for release based on the following criteria:
  - All medications are discontinued two weeks prior to release and animal has maintained good health
  - The animal must maintain normal bloodwork for a minimum of 2-4 weeks after medications are discontinued
  - The animal must have a normal physical examination to include:
    - The animal demonstrates normal swimming and mobility

        - Confirmation that the animal has normal hearing
        - Any external wounds or injuries have healed
        - Resolve of any underlying health conditions, such as pneumonia or gastroenteritis
        - The animal must be eating food on its own, preferably without the presence of humans
    - Team evaluation of the historical, developmental, behavioral, and medical records

- Perform hearing/acoustic testing if the equipment is available (Auditory Evoked Potential-AEP) to determine if the animal has any hearing deficits to determine if they are a candidate for release

- We recommend a meeting with the AFCR3 Alliance team to collectively determine if this animal is suitable for release

- If the animal is determined to be a suitable candidate for release, the following release considerations should then be considered:

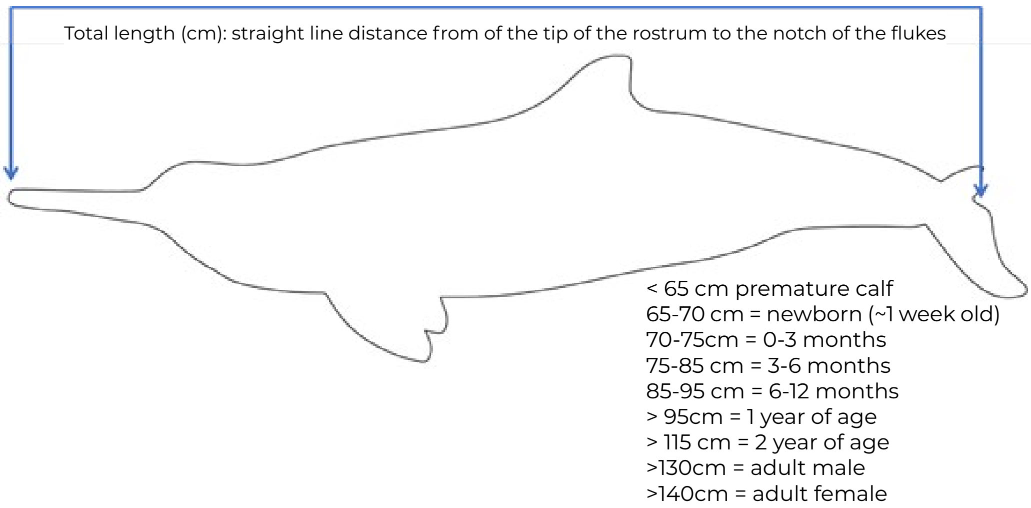
    - Selecting a release location: Ideally release the animal into the home range, genetic stock and social unit within Franciscana management area that stranding occurred
    - Consider application of identification methods prior to release e.g. flipper roto tags, PIT tags, radio tags, satellite tags, freeze branding
    - Release site requirements and recommendations – personalize location according to stranding location, age and sex of individual and time to minimize additional energetic and social demands to maximize foraging success.
    - Maintenance of stock fidelity, proximity of conspecifics, proximity to fishing gear and areas of entanglement risks, timing in relation to breeding seasons and migration activities are also crucial considerations.
    - Post-release monitoring – if available, visual tracking, tagging, standardization of data collection protocols should be considered

Resources:

1. Sweeney, et al. Comparative Survivability of Tursiops Neonates from Three U.S. Institutions for the Decades 1990-1999 and 2000-2009. Aquatic Mammals 2010, 36(3), 248-261, DOI 10.1578/AM.36.3.2010.248
2. Masahiko Kasamatsu, et al. The First Entire Hand-Rearing of Two Newborn Finless Porpoises (Neophocaena asiaeorientalis). IAAAM Proceedings; 2018.
3. Jennifer E. Flower, et al. Neonatal Critical Care and Hand-Rearing of a Bottlenose Dolphin (Tursiops truncatus) Calf. Aquatic Mammals 2018, 44(5), 482-490, DOI 10.1578/AM.44.5.2018.482.
4. Baldassin, P. et al. "Veterinary treatment of an injured wild franciscana dolphin calf (Pontoporia blainvillei , Gervais & d'Orbigny, 1844)." *Latin American Journal of Aquatic Mammals* 6 (2007): 185-187.
5. CRC Marine Mammal Handbook, 3rd edition
6. CRC Marine Mammal Handbook, 2nd edition
7. Marine Mammals Ashore, A field guide for strandings, 2nd edition. Pp 75-112. J Geraci, V Lounsbury. 2005.
8. Caon, et al. Journal of the Marine Biological Association of the United Kingdom, 2008, 88(6), 1099–1101.
9. D. Rodríguez, I. Rivero and R. Bastida. Feeding ecology of the Franciscana (*Pontoporia blainvillei*) in marine and estuarine waters of Argentina. LAJAM 1(1): 77-94, Special Issue 1, 2002.

Appendix 1. Growth Curve-Age estimation for Franciscana dolphins

Measure body length to determine age class. A standard straight length is defined as the measurement of the straight-line distance from the tip of the rostrum to the notch of the flukes.



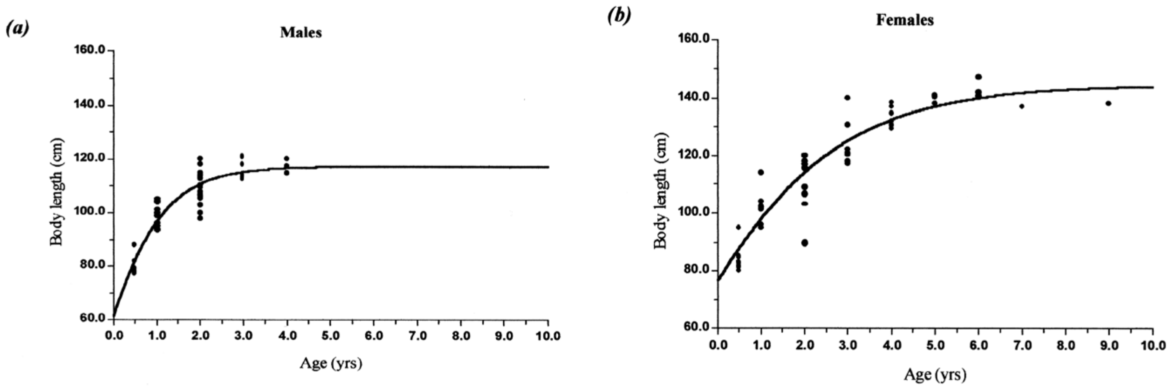
- Sexual maturity, length, and weight varies for males and females but is usually obtained between 2-3 years
- Males:
  - 131cm length and 25-29kg in weight to be classed as an adult
  - Male: average juvenile male weight ~20kg and average adult male weight ~27kg
- Females:
  - 140cm length and 33-34kg in weight to be classed as an adult
  - Average juvenile female weight ~25kg and average adult female weight ~ 33kg

Body length mean (minimum–maximum) for each age-class in males and females of *Pontoporia blainvillei* accidentally captured in gillnets or stranded in northern Rio de Janeiro, Brazil, from 1989 to 1996.

Table previously published by R. M. Arruda Ramos et al. Aquatic Mammals 2000, 26.1, 65–75.

Table 1. Body length mean (minimum–maximum) for each age-class in males and females of *Pontoporia blainvillei* and *Sotalia fluviatilis* accidentally captured in gillnets or stranded in northern Rio de Janeiro, Brazil, from 1989 to 1996.

	Males		Females	
Age	*n*	Length range	*n*	Length range
P. blainvillei				
0	5	81.1 (78.0–88.0)	6	83.2 (74.0–95.0)
1	8	99.4 (94.0–105.0)	7	103.9 (95.0–114.0)
2	18	109.4 (98.0–120.0)	10	111.6 (90.0–120.0)
3	4	116.5 (113.0–121.0)	7	124.2 (117.5–140.0)
4	4	118.1 (115.0–120.0)	6	133.8 (129.5–138.5)
5	—		2	139.5 (138.0–141.0)
≥6	—		5	141.1 (137.0–147.5)
total	39		43	



Scatterplots of length-at-age for 39 males (figure a) and 43 females (figure b) of Pontoporia blainvillei accidentally captured in gillnets or stranded in northern Rio de Janeiro, Brazil, from 1989 to 1996. The solid line represents the predicted growth trajectory from the Gompertz model. Figures previously published by R. M. Arruda Ramos et al. Aquatic Mammals 2000, 26.1, 65–75.

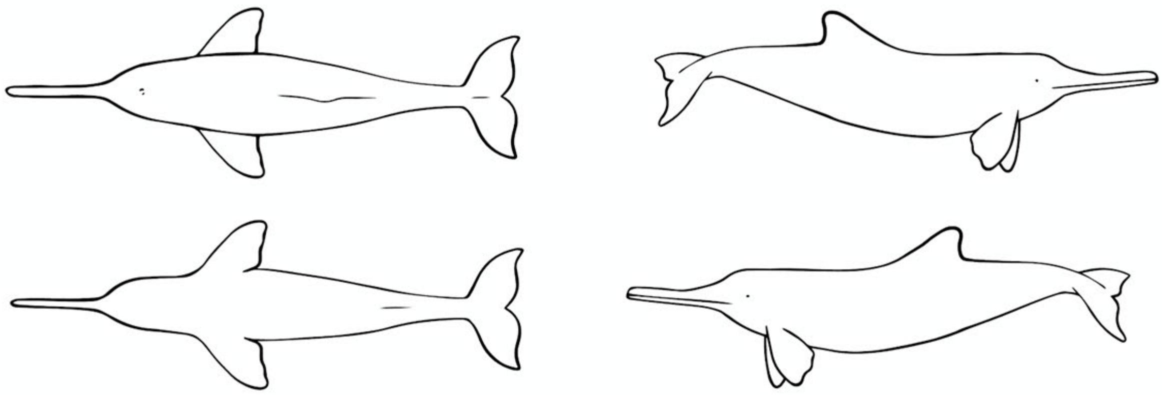
Appendix 2. Animal Monitoring Sheet During Rescue and Transport

ANIMAL MONITORING SHEET - DURING RESCUE AND TRANSPORT													
ANIMAL ID/NAME:													
DATE:													
	On Beach	During transport											
Time:													
Respiratory rate													
Heart Rate													
Temperature													
Behavioral Response:													
Alert													
Weak													
Non-responsive													
Blood glucose:													
Record any treatments given:													
Comments:													

Appendix 3. Franciscana Dolphin (*Pontoporia blainvillei*) Physical Exam Form

Animal Name/ID:		**Date of Exam:**
Weight (kg): Length: (cm) Axial Girth (cm): Sex: ☐ Male ☐ Female	Age class based on length (cm): ☐ < 65 cm premature calf ☐ 65-70 cm = newborn (~1 week old) ☐ 70-75cm = 0-3 months ☐ 75-85 cm = 3-6 months ☐ 85-95 cm = 6-12 months ☐ > 95cm = 1 year of age ☐ > 115 cm: 2 year of age ☐ >130cm = adult male ☐ >140cm = adult female	Body Condition Score (1-5): _____ 1 = emaciated 2 = underweight 3 = ideal 4 = robust 5= obese Mucous membrane color: _____________ Capillary refill time: ___________ (seconds)
Temperature (°C):	Respiratory Rate: breaths/1 min: _____________ breaths/5 min: _____________	Heart Rate: (beats/1 Min): ________ Brad ________Tach ________Mean

System	Abnormal/Normal/Not examined	
Neurological	A/N/NE	Mentation: ☐ Alert ☐ Dull ☐ Stuporous Describe any abnormal findings:
Ophthalmic OD (Right)	A/N/NE	Menace (+ / -) Palpebral (+ / -) Blepharospasm (+ / -) Visual Tracking (+ / -) Corneal lesions (+ / -) Describe any lesions here and draw on the diagram below:
Ophthalmic OS (Left)	A/N/NE	Menace (+ / -) Palpebral (+ / -) Blepharospasm (+ / -) Visual Tracking (A / N) Corneal lesions (+ / -) Describe any lesions here and draw on the diagram below:
Oral/Teeth	A/N/NE	Teeth erupted (+ / -) Gingival hyperplasia (+ / -) Tooth Wear (+ / -) Crown Fractures (+ / -) Describe any lesions:
Respiratory	A/N/NE	Breath quality: ☐ Normal ☐ Shallow ☐ Other: describe: Lung sounds (auscultation): ☐ Normal ☐ Crackles ☐ Wheezes ☐ Rales ☐ Other: describe: Respiratory Mucus: ☐ None ☐ Present; describe: Describe any abnormalities here:
Cardiovascular	A/N/NE	Rhythm: ☐ Normal Sinus Arrhythmia ☐ No Sinus Arrhythmia ☐ Other Arrhythmia Murmur (+ / -) describe any abnormalities here:
Musculoskeletal	A/N/NE	Describe any abnormalities:
Gastrointestinal	A/N/NE	Intestinal (gut) sounds ☐ Present ☐ Not Present Gastric fluid ☐ Normal ☐ Abnormal ☐ Not Collected pH: ______; cytology: ________ ______________________________ Feces ☐ Normal ☐ Abnormal; ☐ Not collected; cytology: ________________________________ ________________________ Vomiting: (+/-); describe appearance and frequency:
Integument	A/N/NE	Describe any skin lesions, rake marks, scars; vibrissae present, etc and draw on the diagram below:
Reproductive	A/N/NE	Umbilicus: ☐ Normal ☐ Abnormal ☐ Open ☐ Healed ☐ Discharge present Genital slit ☐ WNL ☐ Abnormal Vagina/Penis ☐ WNL ☐ Abnormal Mammary (R/L): ☐ WNL ☐ Abnormal



Eyes: Ophthalmic OD (Right) **Ophthalmic OS (Left)**

Comments:____
____
____
____
____
____
____

Veterinarian (Name/Signature): ____

Date: ________________________

Appendix 4. Recipe for Fish Gruel

Recipe Tip: Prepare the gruel fresh daily, and store in the refrigerator until use. Discard any unused gruel after 24 hours.

Franciscana (Juvenile/Adult) fish gruel recipe:

To make 400 mL of fish gruel:
125 ml NaCl or LRS fluids
125 ml filtered water
100-200 grams fish filets
3 mL glicopan
5 grams Glucose
10 ml fish oil = 10 capsules
100 mg taurine
Lecithin: 1 gram
1/16 tablet multivitamin
***20 grams Zoologic 30/52
***30 grams Zoologic 33/40

***If the Zoologic Milk Matrix (PetAg) products are available they can also be added to the formula for additional protein, fat, and calories

Appendix 5. Drug Formulary

Drug Name	Dose	Route	Comments
Amikacin	5-10 mg/kg SID	IM/IV	
Amoxicillin	10-20 mg/kg BID	Oral	
Ampicillin	10-20 mg/kg TID	Oral, IV	
Azithromycin	5-10 mg/kg SID	Oral, IV	
Ceftriaxone	15-20 mg/kg SID	IM/IV	
Cefovecin (Convenia)	8 mg/kg ONCE every 10-14 days	Subcutaneous (SQ), IM	
Ceftiofur (EXCEDE)	6.6 mg/kg ONCE every 5 days	SQ	
Cimetidine	3 mg/kg SID-BID	Oral	
Ciprofloxacin	15 mg/kg BID	Oral	
Doxycycline	5 mg/kg SID 2.5 mg/kg BID	Oral	
Enrofloxacin	5 mg/kg BID	Oral, IV, IM*	*AVOID repeated IM injections with fluoroquinolones can be irritating to the muscles
Fenbendazole	10 mg/kg SID for 3 consecutive days	Oral	
Famotidine	0.5 mg/kg BID	Oral, IV, SQ	
Fluconazole	4-6 mg/kg SID	Oral	
Itraconazole	2.5 mg/kg BID 5 mg/kg SID	Oral	
Levofloxacin	10 mg/kg SID	Oral, IV	
Metronidazole	5 mg/kg BID	Oral	
Ondansetron	0.1 mg/kg SID-BID	Oral, SQ, IM	
Omeprazole	0.2 mg/kg SID	Oral	
Prednisone/Prednisolone	0.05-0.25 mg/kg SID-BID	Oral, IM, IV	
Simethicone	2 mg/kg SID-QID	Oral	Do NOT exceed 8 doses in 1 day
Sucralfate	5-10 mg/kg SID-TID	Oral	

EMERGENCY DRUGS AND SEDATION DRUGS TO HAVE AVAILABLE:

Franciscana Emergency Drug Doses: SMALL JUVENILE						
Animal ID/Name		Weight (kg):	10	Date:		
Drug	**Route**	**Dosage (mg/kg)**	**Conc. (mg/ml)**	**Amount (mg)**	**Amount (mL)**	**Indications for use**
Midazolam	IM	0.06	5	0.6	**0.12**	Sedation, anxiety, seizures
Diazepam	PO, IM	0.1	5	1.0	**0.2**	Sedation, anxiety, seizures
Flumazenil	IM, IV	0.01	0.1	0.1	**1.0**	Reversal drug to reverse effects of diazepam or midazolam
Emergency Drugs	**Route**	**Dosage (mg/kg)**	**Conc. (mg/ml)**	**Amount (mg)**	**Amount (mL)**	**Indications for use**
Atropine	IM, IV	0.02	0.4	0.2	**0.5**	Bradycardia
Calcium Gluconate	IM, or IV slowly	10	100	100.0	**1.0**	Hypocalcemia, muscle tremors/weakenss
Diphenhydramine	IV,IM	1	50	10.0	**0.2**	Allergic reaction
Doxapram	IM, IV, IT, sublingual	1	20	10.0	**0.5**	Respiratory stimulant
Epinephrine	IV, IT, IC	0.1	1	1.0	**1.0**	Cardiac resuscitation
Furosemide	IM, IV	1	10	10.0	**1.0**	Pulmonary edema
Lidocaine	IV slowly	2	20	20.0	**1.0**	Ventricular arrhythmia
Sodium Bicarb (mEq/ml)	IV	0.5	1	5.0	**5.0**	Acidosis/Hyperkalemia
Solu-Medrol (methylprednisolone sodium succinate)*	IM, SQ, IV	1	125	10.0	**0.08**	Shock, allergic reaction
Dexamethasone SP (sodium phosphate)*	IM, SQ, IV	0.5	4	5.00	**1.25**	Shock, allergic reaction
*These steroid doses are for emergency resuscitation. Give a lower dose if using as an anti-inflammatory (0.1-0.5 mg/kg)						

Franciscana Emergency Drug Doses: JUVENILE DOLPHIN						
Animal ID/Name		Weight (kg):	20	Date:		
Drug	**Route**	**Dosage (mg/kg)**	**Conc. (mg/ml)**	**Amount (mg)**	**Amount (mL)**	**Indications for use**
Midazolam	IM	0.06	5	1.2	**0.24**	Sedation, anxiety, seizures
Diazepam	PO, IM	0.1	5	2.0	**0.4**	Sedation, anxiety, seizures
Flumazenil	IM, IV	0.01	0.1	0.2	**2.0**	Reversal drug to reverse effects of diazepam or midazolam
Emergency Drugs	**Route**	**Dosage (mg/kg)**	**Conc. (mg/ml)**	**Amount (mg)**	**Amount (mL)**	**Indications for use**
Atropine	IM, IV	0.02	0.4	0.4	**1.0**	Bradycardia
Calcium Gluconate	IM, or IV slowly	10	100	200.0	**2.0**	Hypocalcemia, muscle tremors/weakenss
Diphenhydramine	IV,IM	1	50	20.0	**0.4**	Allergic reaction
Doxapram	IM, IV, IT, sublingual	1	20	20.0	**1.0**	Respiratory stimulant
Epinephrine	IV, IT, IC	0.1	1	2.0	**2.0**	Cardiac resuscitation
Furosemide	IM, IV	1	10	20.0	**2.0**	Pulmonary edema
Lidocaine	IV slowly	2	20	40.0	**2.0**	Ventricular arrhythmia
Sodium Bicarb (mEq/ml)	IV	0.5	1	10.0	**10.0**	Acidosis/Hyperkalemia
Solu-Medrol (methylprednisolone sodium succinate)*	IM, SQ, IV	1	125	20.0	**0.16**	Shock, allergic reaction
Dexamethasone SP (sodium phosphate)*	IM, SQ, IV	0.5	4	10.00	**2.50**	Shock, allergic reaction
*These steroid doses are for emergency resuscitation. Give a lower dose if using as an anti-inflammatory (0.1-0.5 mg/kg)						

Franciscana Emergency Drug Doses: ADULT DOLPHIN						
Animal ID/Name		Weight (kg):	30	Date:		
Drug	**Route**	**Dosage (mg/kg)**	**Conc. (mg/ml)**	**Amount (mg)**	**Amount (mL)**	**Indications for use**
Midazolam	IM	0.06	5	1.8	**0.36**	Sedation, anxiety, seizures
Diazepam	PO, IM	0.1	5	3.0	**0.6**	Sedation, anxiety, seizures
Flumazenil	IM, IV	0.01	0.1	0.3	**3.0**	Reversal drug to reverse effects of diazepam or midazolam
Emergency Drugs	**Route**	**Dosage (mg/kg)**	**Conc. (mg/ml)**	**Amount (mg)**	**Amount (mL)**	**Indications for use**
Atropine	IM, IV	0.02	0.4	0.6	**1.5**	Bradycardia
Calcium Gluconate	IM, or IV slowly	10	100	300.0	**3.0**	Hypocalcemia, muscle tremors/weakenss
Diphenhydramine	IV,IM	1	50	30.0	**0.6**	Allergic reaction
Doxapram	IM, IV, IT, sublingual	1	20	30.0	**1.5**	Respiratory stimulant
Epinephrine	IV, IT, IC	0.1	1	3.0	**3.0**	Cardiac resuscitation
Furosemide	IM, IV	1	10	30.0	**3.0**	Pulmonary edema
Lidocaine	IV slowly	2	20	60.0	**3.0**	Ventricular arrhythmia
Sodium Bicarb (mEq/ml)	IV	0.5	1	15.0	**15.0**	Acidosis/Hyperkalemia
Solu-Medrol (methylprednisolone sodium succinate)*	IM, SQ, IV	1	125	30.0	**0.24**	Shock, allergic reaction
Dexamethasone SP (sodium phosphate)*	IM, SQ, IV	0.5	4	15.00	**3.75**	Shock, allergic reaction
*These steroid doses are for emergency resuscitation. Give a lower dose if using as an anti-inflammatory (0.1-0.5 mg/kg)						

Appendix 6. Daily Monitoring Sheet

Date: ______________________ Animal ID: ________________________

Time	Respiratory Rate	Describe behavior (swim patterns, vomiting, crunching, arching, independence)	Formula concentration	Formula volume (ml)	Defecation: Describe feces frequency, color, consistency	Calories intake (total)	Calories intake (kcal/kg/ day)	Weight (kg)	Weight Gain (kg)	Length (cm)

Appendix 7. Water Quality Monitoring Data Sheet

- Please monitor the following water quality parameters **every day** and record on the form
  - Water temp: 25-28 Celsius
  - Salinity: 20-35 ppt
  - pH: 7.2-8.4
  - Ammonia < 2 ppm
  - If chemical filtration is used, these chemicals will need to measured daily as well (i.e. chlorine, ozone, etc)

- Please monitor the bacterial coliform count **once per week** and record on the form
  - coliform count must be < 1,000 MPN (most probable number)
  - **if the coliform count is > 1,000 = increased fecal bacteria in the water, this requires a pool cleaning and a water change of at least 25% then repeat the test

Date	Time	Temperature (celcius)	Salinity (ppt)	pH	Ammonia (ppm)	Coliform count	Comments

Appendix 8. Stomach contents free-ranging Franciscana dolphins – based on length and age

- 12 common species of fish have been found in stomach contents of Franciscana from Brazil to Uruguay including:
  - Five types of *sciaenidae* (drums or croakers)
    - Paralonchurus brasiliensis
    - Cynoscion striatus
    - Macrodon ancylodon
    - Micropogonias furnieri
    - Umbrina canosai
  - Two types of *Engraulidae* (anchovy)
    - Engraulis anchoita
    - Anchoa marinii
  - *Porichthys porosissimus* (toadfish)
  - *Urophycis lepturus* (Brazilian codling)
  - *Peprilus paru* (harvestfish / butter fish)
  - *Trichiurus lepturus* (largehead hairtail)
  - *Trachurus lathami* (jack mackerel)

- Squid beaks (*Loligo sanpaulensis*) were also common

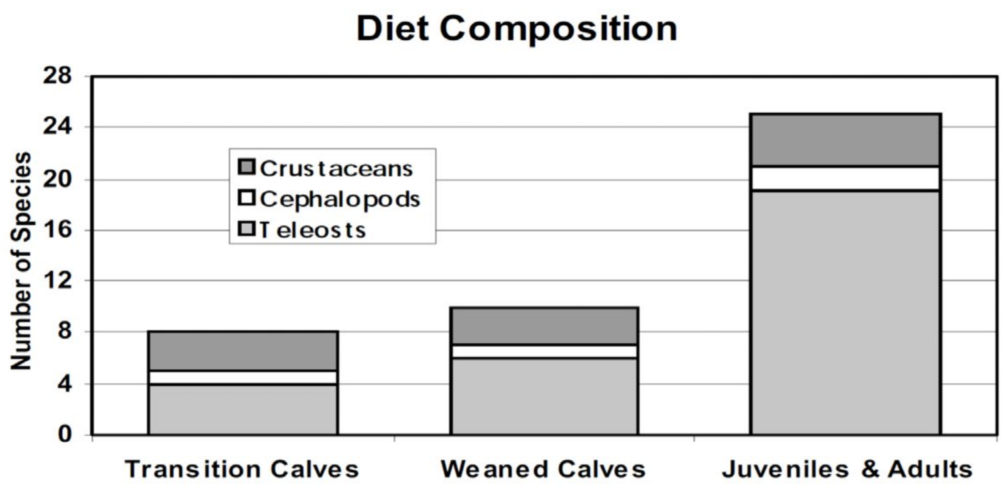
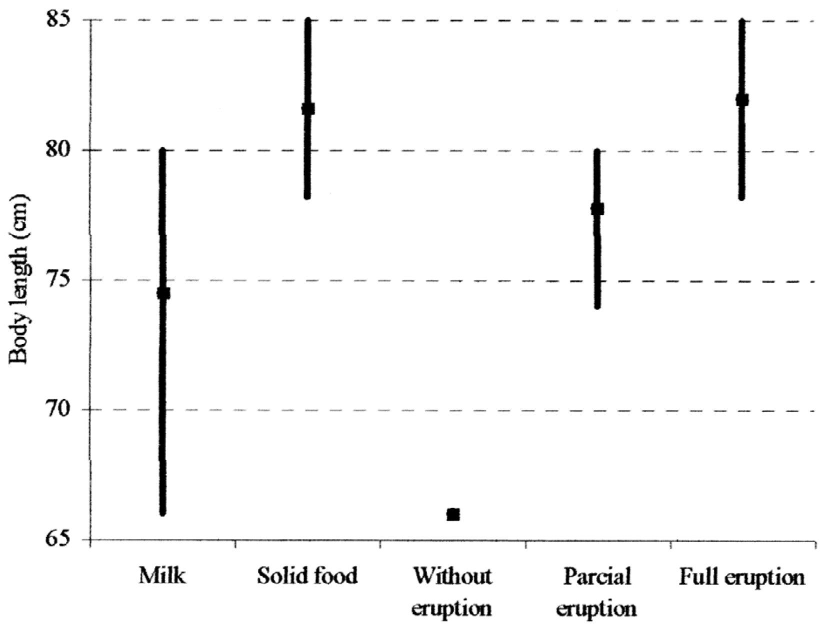


Figure 5. Number of prey species taken by franciscanas of different ontogenic categories.

Number of prey species taken by Franciscanas of different ontogenic categories. Figure previously published by D.RODRÍGUEZ, L.RIVERO and R.BASTIDA. LAJAM 1(1): 77-94, Special Issue 1, 2002



Analyses of stomach content (milk or solid food) and three tooth eruption stages (without, parcial and full eruption) to body length of calves (newborn and 0.5 GLG) of Pontoporia blainvillei accidentally captured in gillnets or stranded in northern Rio de Janeiro, Brazil, from 1989 to 1996. All calves over 80.0 cm presented some alimentary remains (i.e., fish otoliths) in the stomach. Calves between 78.0 and 80.0 cm length had milk (n=2) or food solid (n=2). Figure previously published by Ramos, et al. Aquatic Mammals 2000, 26.1, 65–75.

Absolute number (n=), numerical abundance (N%), frequency of occurrence (FO%), biomass (W%), absolute (IRI) and percentage Index of Relative Importance (IRI%) of prey of franciscanas from northern Argentina. Table previously published by D.RODRÍGUEZ, L.RIVERO and R.BASTIDA. LAJAM 1(1): 77-94, Special Issue 1, 2002

PREY ITEMS	'TRANSITION' CALVES (n=14)			WEANED CALVES (n=14)			POOLED JUVENILES & ADULTS (n=60)					
	n=	N%	FO%	n=	N%	FO%	n=	N%	FO%	W%	IRI	IRI%
TELEOSTS	**36**	**10.4**	**71.4**	**263**	**90.4**	**100.0**	**2844**	**80.5**	**96.9**	**72.0**	**14780.3**	**92.2**
Micropogonias furnieri	2	0.6	25.0	133	45.7	50.0	500	14.1	40.9	24.3	1574.3	20.4
Cynoscion guatucupa	29	8.4	50.0	69	23.7	83.3	1637	46.3	60.6	23.8	4250.8	55.2
Odonthestes argentinensis				37	12.7	16.7	198	5.6	13.6	6.7	167.5	2.2
Macrodon ancylodon							17	0.5	10.6	6.7	76.7	1.0
Paralonchurus brasiliensis	4	1.2	12.5	16	5.5	33.3	127	3.6	19.7	2.3	116.9	1.5
Urophycis brasiliensis				3	1.0	33.3	116	3.3	24.2	7.4	258.9	3.4
Mugil platana							9	0.3	7.6	0.1	2.5	0.0
Engraulis anchoita				5	1.7	33.3	28	0.8	24.2	0.4	29.8	0.4
Stromateus brasiliensis							7	0.2	3.0	0.2	1.1	0.0
Umbrina canosai							125	3.5	15.2			
Lycengraulis olidus							4	0.1	4.5			
Pomatomus saltatrix							2	0.1	3.0			
Ramnogaster arcuata							9	0.3	3.0			
Percophis brasiliensis							1	0.1	1.5			
Sparus pagrus							11	0.3	7.6			
Trachurus lathami							49	1.4	1.5			
Pogonias cromis							2	0.1	1.5			
Raneya fluminensis							1	0.1	1.5			
Anchoa marini							1	0.1	1.5			
Leptonotus blanvillanus	1	1.0										
CEPHALOPODS	**1**	**0.3**	**12.5**	**16**	**5.5**	**16.7**	**443**	**12.5**	**30.8**	**28.0**	**1248.3**	**7.8**
Loligo sanpaulensis	1	0.3	12.5	16	5.5	16.7	441	12.5	30.3	28.0	1227.5	15.9
Octopus tehuelchus							2	0.1	1.5			
CRUSTACEANS	**181**	**52.5**	**85.7**	**9**	**3.1**	**50.0**	**238**	**6.7**	**49.2**			
Artemesia longinaris	3	0.9	12.5	1	0.3	16.7	197	5.6	28.8			
Peisos petrunkievitchi	9	2.6	12.5	7	2.4	33.3	24	0.7	15.2			
Peneidae	4	1.2	25.0	1	0.3	16.7	7	0.2	10.6			
Neomysis americana	135	39.1	25.0				6	0.2	1.5			
Pleoticus muelleri							4	0.1	1.5			
Calanoid Copepods	30	8.7	25.0									
OTHERS	**128**	**37.1**	**25.0**	**3**			**8**					
Nereid Polychaetes	128	37.1	25.0	3	1.0	50.0	8	0.2	10.6			
TOTAL	**346**			**291**			**3533**					

The diet of both transition and weaned calves was found to be less species rich than older animals. The solid diet of transition calves from both habitats was more similar to the diet of estuarine juveniles and adults, suggesting certain dependence of calves on brackish habitats during the transition to solid feeding. An important prey species during this period was the mysid *Neomysis americana* (opossum shrimp), which is very abundant in Bahía Samborombón and also a major constituent of other species' diet in the Rio de la Plata estuary, such as *Micropogonias furnieri* (white-mouthed croaker) (FO%=55%, Sánchez et al., 1991) and M. ancylodon (FO%=88.8%, Leta, 1987). The Argentine red shrimp, Pleoticus muelleri, and A. longinaris are also frequently eaten by juvenile franciscanas in southern Brazil (Bassoi, 1997; Bassoi et al., 2000) and a high incidence of euphausiids characterise the transition diet of harbor porpoises from the Gulf of Maine (Smith and Read, 1982; Gannon et al., 1998). Smith and Read (1992) suggested that these calves eat crustaceans while mothers are feeding on euphausiid predators, a strategy that is likely to occur with Pontoporia and Neomysis. In contrast, the prey species composition of post-weaning calves from both habitats resembles that of juvenile and adult franciscanas from marine habitats. (D.RODRÍGUEZ, L.RIVERO and R.BASTIDA. LAJAM 1(1): 77-94, Special Issue 1, 2002)

Appendix 9. Common disease/conditions in a juvenile/adult stranded Franciscana

- **Pneumonia** is common in stranded cetaceans. Ultrasound examination of the lungs can provide an indication of mild, moderate or severe lung disease. Assessment of respiratory rate, character, auscultation and blow assessment for malodor or mucus can all aid diagnosis. Combining with an azole if fungal pneumonia is suspected can also be beneficial. Serology or PCR can be useful in diagnosing fungal pneumonia as blowhole swabs are frequently inaccurate.

- **Gastrointestinal disease** - stranded cetaceans depending on the underlying cause could have potentially not eaten in some time. Gastrointestinal disease can be confirmed via abdominal ultrasound, gastric fluid cytology, gastroscopy and fecal cytology.

- A high burden of gastrointestinal parasites is likely in a debilitated wild cetacean. De-worming protocols are available however caution should be taken to reduce a high load of deceased parasites causing more gastrointestinal issues. Animal stability is paramount prior to deworming. Common parasites confirmed in Franciscana dolphins include ectoparasites *Xenobalanus globicipitis* found on the trailing edges of the fluke, and *Cirolana* found in blowholes and stomachs.

- Endoparasites include nematode *Contracecum* and acanthocephalan *Polymorphus* both parasites have been found in the stomach (Brownell 1975). Several animals have had ulcerated stomachs on necropsy exam as a result of these parasites. Larval cestodes (*Phyloobothrium* and *Monorygma*) have not been observed in Franciscana.

- **Skin disease** – Entanglement wounds and lacerations are likely to be present. Deeper wounds with infection will require topical cleaning and systemic antibiotics. Changes in water temperature and salinity can cause rapid alterations in cetacean skin therefore close monitoring during the initial days of rehabilitation is important. This can provide an indication of environmental quality. Photographs of skin lesions are important to monitor progress. Systemic medication can be used to treat but ensuring good water quality is of paramount importance.

- **Auditory and Visual Activity -** Auditory dysfunction, involving production or reception of typical sounds or signals occurring in the wild, may indicate active disease, permanent injury, or degenerative changes associated with aging. Evaluators may suspect that a cetacean has compromised auditory function if it appears to have difficulty locating prey items or various objects via echolocation or if it minimally responds to novel noises.

-

Appendix 10. Ethogram Franciscana *(Pontoporia blainvillei)*

Rationale

In order to be able to assess the overall condition of an animal and its development in the context of rehabilitation, behavioural data are indispensable. Behavioural data can not only document progress in the rehabilitation process, more importantly, certain behaviours that indicate a change of the animal's health, e.g., that the animal is weakened, can be detected early, so that appropriate action can be taken. Also, well designed and conducted behavioural study can bring to light certain behaviours that are overlooked in everyday life.

The ethogram

In order to describe a behaviour pattern, it is recommended to distinguish between two important types of behavioural activity which lie at opposite ends of a continuum. On the one hand we have behavioural **events**, that are behaviour patterns of relatively short duration, such as breathing, vocalizations, etc. The salient feature of events is their frequency, i.e. how many times the animal breathes per minute. The second category comprises behavioural **states**, that are of relatively long duration. Their salient feature is duration, i.e. how many minutes is the animal swimming at relatively fast speed.

The ethogram contains codes and descriptions for activity states and for behavioural event types. This ethogram was created based on observations of single individuals and therefore only includes solitary behaviors.

The activity states (salient feature duration) comprise:

Code	Name	Description	Interpretation regarding animals condition
SW	Swimming	Locomotion by up and down movement of the fluke	Neutral
SW-Sl	Swimming slowly	The animal moves slowly	Depends on the context / might signal weakness
SW-Fa	Swimming fast	The animal is moving fast	Indicates good condition
SW-UP	Swimming Upside Down	The animal is swimming belly up	Neutral
SW-SW	Swimming sideway	The animal swims slowly and sideways, the body is tilted sideways by approx. 45°.	Depends on the context / might signal weakness
SW-Dis	Discontinuous swimming	The animal swims erratically without an even rhythm	Depends on the context / might signal weakness
SW-Gr	Swimming on the ground	The animal swims submerged and touches with his body the ground of the pool	Neutral
BB	Bent Body	The animal's body is somehow bent upwards.	indicates weakness
Hd-SC	Head scanning	The animal moves the head from right to left on a horizontal plane, no object nearby	Indicates good condition
APP	Approach	The animal swims directly towards caretaker (e.g., as soon as he puts hand into the water)	Indicates good condition
REA	Rest active	The animal remains on the same place but shows some flukes strokes	Neutral
REP	Rest passive	The animal remains motionless on the same place for some time, lies on the floor or floats at the water surface	Neutral
EXP	Exploration	The animal swims slowly, moving its head from left to right in relation to an object in front of it.	Indicates good condition
TO	Touch object	The animal touches intentionally an object lying in the water with its beak	Indicates good condition
AO	Avoiding object	The animal avoids objects that are in its swimming lane	Indicates good condition
DI	Diving	The animal leaves the surface and dives down. No bubbles are visible	Neutral
Acc	Abrupt Acceleration	The animal shows an abrupt acceleration	Indicates good condition
SWCh	Change of swimming direction	The animal shows a fast change in swimming direction	Indicates good condition
BU	Bubble	The animal produces a considerable amount of bubbles under water	Neutral
RO	Rolling	The animal turns around its own longitudinal axis	Neutral

IMPORTANT: For the Category Swimming a modifier can be applied to distinguish Clockwise Swimming (CW) from Anticlockwise Swimming (ACW)

For example:
SW-CW
SW-ACW

Event types (salient feature frequency) comprise:

Code	Name	Description	Interpretation regarding animals condition
BR	Breathing	Animal comes to the surface and breathes in and out	Number of breaths/minute can give an indication of the animal's condition.
BU	Bumping	Animal swims headfirst into the pool wall	Depends on the context / might signal weakness
CH	Chuff	The animal expels air abruptly	Depends on the context / might signal weakness / repeated chuffs should be monitored.
HJ	Head jerking	The animal raises its head jerkily when breathing	Indicates weakness
MO	Mouth open	Upper and lower jaw separated in a non-feeding situation	Neutral
IN	Ingest	The animal takes a fish and swallows it	Indicates good condition
EX	Excretion	Defecating	Frequency of defecation, color, and consistency

Methods

Regarding sampling rules: in this special case (one individual) a focal sampling method and a continuous recording rule (or all-occurrences recording) is recommended. In practice: we suggest performing sessions of 30 Minutes duration three times a day.

Behavior Recording Sheet			
Start Time Hs/Min/Sec	**Behavior State**	**Behavior Events**	**Observations**

Alliance for Franciscana-Dolphin
Conservation Research, Rescue and Rehabilitation AFCR3

National Marine Mammal Foundation
yaqu pacha
Tiergarten Nürnberg
Dolphin Quest
Integrated Conservation Planning for Cetaceans
IUCN SSC

Aiuká
Aquarium Mar del Plata
Karumbé www.karumbe.org
AquaMarina
Fundacion Mundo Marino
Mundo Marino Creando lazos para conservar
R3 Animal salvando vidas
FURG Universidade Federal do Rio Grande
yaqu pacha Uruguay