Pyothorax in a Cat

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## Introduction

This case report describes an eight-year-old male neutered domestic shorthair cat that presented for lethargy and inappetence. The cat was diagnosed with pyothorax using thoracic radiographs, ultrasound and cytology of the pleural effusion. He was medically managed with the placement of bilateral thoracostomy tubes, intravenous antibiotics and analgesics. He was discharged from the hospital after seven days on oral antibiotics and made a clinical recovery based on thoracic radiographs taken seven days after discharge as well as owner history and physical examination one month after discharge.

Pyothorax is the accumulation of a purulent exudate in the pleural space, which is the potential space between the lungs, mediastinum, diaphragm, and thoracic wall. [1,2,3] A small amount of fluid is normally contained within the pleural space, which allows structures to move freely during respiration. [3,4] Starling's forces govern pleural fluid formation and drainage, and pleural effusion can develop when disease processes alter the normal fluid dynamics within the pleural space. [2,3] Inflammatory conditions caused by pyothorax result in increases in capillary permeability and obstruction of lymphatic drainage due to the release of chemical mediators. [1,2,3,4] These lead to an influx of fluid, protein, and cells into the pleural space. Continuous fluid accumulation eventually results in pulmonary atelectasis and hypoventilation. [2]

Pyothorax can arise from hematogenous or lymphatic origin, via extension from an adjacent structure, or via direct inoculation into the pleural space. [1,5] Bacteria can enter the pleural space from compromised lung parenchyma, trachea, bronchus, esophagus or thoracic wall. [1] It was commonly postulated in the veterinary literature that feline pyothorax was caused by penetrating bite wounds and the resultant inoculation of oral flora into the pleural space. [4,6,7,8] This was supported by the fact that the most commonly found microbes in pyothorax are obligate and facultative anaerobes also present in the feline oropharynx, which are similar to those found in subcutaneous bite abscesses. [5,9] In addition, cats from multi-cat households and cats with frequent access to the outdoors appear over-represented. [6,8] In one report, cats with pyothorax were 3.8 times more likely to originate from multi-cat households, and the authors proposed that inter-cat aggression was responsible for small, unnoticed bite wounds. [8]

By the time cats presented to the hospital, however, no evidence of a thoracic wound was found in the majority of cases, and the cause of the infection was usually not determined antemortem. [5,8,10] A recent history of a bite or other external wound was only documented in 4%-14.5% of cases. [5,8]

A recent report on feline pyothorax offers an alternative explanation for the etiology: it is the result of aspiration of oropharyngeal flora and subsequent colonization of the lower respiratory tract, with direct extension of infection from the bronchi and lungs into the pleural space. [5] In a retrospective study reviewing 27 cases of feline pyothorax in Australia, a cause for the development of pyothorax was found in 18 of 27 cats (67%), and the mechanism of infection was identified as extension of infection from an adjacent intrathoracic structure in 16 of the 18 cats. [5] Identified causes included upper respiratory infection in eight cats (seven of which were suspected to have bronchopneumonia and pyothorax as a sequelae), pneumonia in four cats, perioperative aspiration of oropharyngeal flora resulting in bronchopneumonia and pyothorax in two cats, and bronchopneumonia and pyothorax secondary to parasitic migration in two cats. [5] The author speculated that a recent viral upper respiratory infection may temporarily impair the mucociliary clearance of lower respiratory secretions in cats, a condition that has been described in humans and horses, which may predispose affected cats to pleuropneumonia. [5] Cats from multi-cat households may be over-represented for pyothorax due to increased risks of developing a contagious viral upper respiratory tract infection rather than due to inter-cat aggression. [5]

Cats presenting with pyothorax are generally young, with the mean and median age at presentation approximately five years old, although cats of any age may be affected. [5,6,8,10] There is no gender, age or breed predisposition, and intact males are not over-represented. [5,6,8,11] Vaccine status does not affect risk. [8] The most common owner complaints at presentation include partial or complete anorexia, dyspnea, and lethargy. [5,6,8,10,11,12,13] Less frequent complaints include hypersalivation, weight loss, signs of pain, and ocular or nasal discharge. [8,12] The duration of clinical signs is generally one to two weeks, but signs may be present for several months. [5,6,10]

The most common physical examination findings include increased respiratory effort, a restrictive or paradoxical pattern of breathing, abnormal lung sounds, muffled heart sounds, tachypnea, pyrexia, poor body condition, and dehydration. [5,6,8,10,11,12,13,14] The observation of a restrictive breathing pattern, in which the abdominal viscera is

aspirated toward the chest during inspiration, can provide important evidence for the presence of pleural space disease in cats. In a retrospective study of 195 dogs and 194 cats with a diagnosis of dyspnea, restrictive or paradoxical breathing was a sensitive sign associated with pleural space disease – 90% of cats with this breathing pattern had pleural space disease. [15] Less common physical examination findings in cats with pyothorax include diminished pulse quality and mental dullness. [8] In one study, 15% of cats had signs of an upper respiratory infection at presentation and an additional 15% of owners reported a history of a recent upper respiratory infection. [5]

Differential diagnoses for pleural effusion other than pyothorax include left or right congestive heart failure, neoplastic effusion (mediastinal, bronchopulmonary or primary pleural neoplasia), effusive feline infectious peritonitis, trauma, coagulopathy, peritoneopericardial diaphragmatic hernia, traumatic diaphragmatic hernia, pulmonary thromboembolism, lung lobe torsion, pancreatitis, glomerulonephropathies and *Aelurostrongylus abstrusus* infection. [2,14,16,17]

The most important elements in the diagnosis of pyothorax are imaging (thoracic radiographs and thoracic ultrasound) and pleural fluid analysis (gross appearance, cytological examination, and anaerobic and aerobic culture and sensitivity). Thoracic radiographs can identify the amount of pleural effusion present, whether it is bilateral or unilateral, and may help identify the presence of pulmonary or mediastinal masses (although masses may not be visible until the pleural effusion has been evacuated). [18] The majority of cats have bilateral pleural effusion (76-90%)[5,6,8,10], with the remainder exhibiting unilateral pleural effusion. If a cat has severe respiratory distress, a dorsoventral radiograph may be diagnostic and can avoid the stress of further patient handling. [18] Typical changes observed on radiographs in cats with pyothorax include retraction of pulmonary lobar borders from the thoracic wall, pulmonary atelectasis, accentuation of the lobar edges and interlobar fissures, blurred cardiac silhouette and diaphragmatic border, appearance of a widened mediastinum, and collapse of lung lobes. [1,4,6,10,11]

Thoracic ultrasonography is helpful in identifying pleural effusion and can help differentiate pyothorax, which is an exudate, from other forms of pleural effusion, which are often transudates. Transudates typically have an anechoic appearance on ultrasound, while purulent pleural effusion may be hyperechoic or have a complex echogenicity. [11,12] In addition, pyothorax effusion may be septate due to fibrinous tags between the parietal and visceral pleura. [10,11,12]

Thoracic ultrasonography can also be helpful to identify consolidated lung lobes or masses, cardiac enlargement, mediastinal masses, and abscessed or neoplastic lung nodules, as well as to find small amounts of effusion or the area with maximum effusion. [1,10] This can be useful in selecting a site for diagnostic thoracocentesis.

Gross examination of pleural fluid obtained from thoracocentesis can support the diagnosis of pyothorax. Septic exudates are typically turbid to opaque, often contain flocculent material, and range in color from cream to pink, green-tinged or red. [1,10,11] The most noticeable feature, found in 80% of pyothorax cases [18], is the distinctive foul odor of mixed anaerobic infections, similar to those noted during drainage of cat bite abscesses. [1,11,19] The lack of a characteristic odor does not rule out a pyothorax, however, but may indicate the presence of an unusual pathogen (e.g. aerobes, yeast, or *Mycoplasma* spp). [11]

In-house fluid analysis and cytology can help confirm a diagnosis of pyothorax while laboratory results are pending. Fluid associated with infection typically has a high protein content (greater than 3.0 g/dL) and a specific gravity of at least 1.025. [1,2,11] In-house cytology of pleural fluid, using Wright-Giemsa stained smears, shows a very a high cell count with neutrophils as the predominant cell type (more than 85% of total nucleated cell count). [10,11,13] In addition, large numbers of pleomorphic, intracellular and/or extracellular bacteria are also observed, which may be filamentous, cocci or rods. [10,11,12,19]

Aerobic and anaerobic cultures should be submitted prior to treatment with antibiotics, as pretreatment might result in an absence of growth on the culture. [8,10] It is important to submit both aerobic and anaerobic cultures, as both bacterial forms are frequently present in feline pyothorax infections. [5,8,9,13,19] In a study of 47 cats with pyothorax, obligate anaerobic bacteria were isolated from 89% of feline pleural effusion samples, and a mixture of obligate anaerobic and facultative bacteria were isolated from 44% of samples. [9] In another study of 26 cats with pyothorax, 87.5% of cats had a mixture of two to four different bacterial species, and obligate anaerobes were the most common isolates (70%), followed by facultative anaerobes (22.5%). [13] The most common aerobic bacteria isolated are non-enteric *Pasteurella* spp (40-48%) and *Actinomyces* spp (15%). [8,9] The most common obligate anaerobic bacterial isolated from pleural fluid samples in cats are *Peptostreptococcus anaerobius* (20%), *Clostridium* spp (1-38%), *Fusobacterium* spp (17%), and *Bacterioides* spp (15-24%) [8,9] The majority of cats (63-65%) have multiple bacterial

species cultured from the pleural effusion.[5,8,9,13] On average, 2.1 species of obligate anaerobic bacteria and 1.2 species of aerobic bacteria were isolated from the pleural fluid sample of cats.[9]

Hematology, serum biochemistry, complete urinalysis and retroviral testing should be part of the minimum database to help direct medical management of the patient. Neutrophilic inflammation, often with a left shift, is the most common hematologic finding (36-73%), and toxic changes to neutrophils are also commonly observed.[6,8,11,13] In one study, surviving cats typically had a higher neutrophil count than non-survivors (mean of surviving cats 31,610 ± 23,111 cells/μL compared to mean of non-surviving cats 12,793 ± 6,204 cells/μL).[8] The authors hypothesized that the lower neutrophil count in cats that died was probably secondary to severe sepsis and sequestration of neutrophils in the pleural space of cats with more severe disease.[8]

Mild biochemical abnormalities in cats with pyothorax are common but not specific. The most common abnormalities on serum biochemistry include hypoalbuminemia, hyperglobulinemia, hypoglycemia or hyperglycemia, hyponatremia, hypochloremia, hypocalcemia, and mild elevations of total serum bilirubin concentration and alanine transaminase activity, possibly secondary to hypoxia-induced damage to hepatocytes due to poor perfusion from hypovolemia and sepsis.[1,6,8,10,12,13,20] Positive retroviral status is not a common finding in cats presenting with pyothorax, and there does not appear to be a significant association between retroviral infection and pyothorax.[6,18] In one American study of 80 cats presenting with pyothorax, 4% were feline leukemia virus (FeLV) positive, and 3% were feline immunodeficiency virus (FIV) positive[8] compared to 2.3% FeLV positive and 2.5% FIV positive in a recent cross-sectional study of more than 18,000 cats in North America.[21]

Cats presenting with pyothorax are often critically ill and gentle handling to avoid stress and stabilization while collecting diagnostic information is important. If the hemoglobin saturation is less than 90% in cats breathing room air, supplemental oxygen should be provided.[2,11,14] An oxygen chamber is the least stressful way to provide supplemental oxygen, but flow-by or a facemask are also acceptable techniques.[2] Correction of hypothermia, hypotension, hypoglycemia, hypovolemia and electrolyte imbalances are also important to patient stabilization.[11] Therapeutic needle thoracocentesis with the patient positioned in sternal recumbency can improve respiratory distress, but the effusion may be too thick or loculated to remove large volumes using this technique.[1]

Placement of large bore (14 to 16 FR) bilateral indwelling thoracostomy tubes under general anesthesia or sedation is the recommended treatment of pyothorax. [1,4,5,8,11,22] Repeated needle thoracocentesis is only recommended when the volume of effusion is small or when euthanasia is the only alternative, as repeated thoracocentesis is an inefficient method for complete thoracic drainage and is associated with higher reported mortality rates. [1,5,11,22] Thoracostomy tubes should be placed bilaterally unless radiographs show that the effusion is clearly unilateral. [1,4,11] Septic pleural effusion is seldom isolated to a single hemithorax. [1,4] There is controversy whether the mediastinum in cats is complete or whether fenestrations allowing communication between the two sides of the thoracic cavity are present. [1,4] Even if fenestrations are present, it is likely that they become plugged under inflammatory conditions, limiting the free flow of fluid across the mediastinum. [1] In addition, thoracostomy tubes have a high rate of complications (58% in one study) [5] and bilateral tubes provide redundancy if one tube fails. Reported complications include failure of drainage due to kinking or poor positioning, subcutaneous edema, subcutaneous abscesses, pneumothorax, and radiographic evidence of poor drain positioning. [5,10]

A recent study evaluated the use of small-bore wire-guided thoracic drains for managing pleural space disease in ten dogs and ten cats. [23] The advantages of using the modified Seldinger technique for thoracic drain placement include the use of sedation rather than general anesthesia in critically ill animals, rapid placement time (less than ten minutes for most drains), minimal clinician training, and minor reported complications, including pneumothorax, kinking and malpositioning. [23] In this study, no drains required removal due to obstruction (18/29 drains were placed for the management of pyothorax). [23] In addition, these drains subjectively appeared more comfortable for the animals, and animals with small-bore chest drains required less opioid analgesia than animals managed with large bore chest drains. [23]

After drain placement, radiographs should be obtained to assess their positioning. [22] Intermittent suction may be used to manage thoracostomy tubes (two to six times per day). Continuous suction is more expensive than intermittent suction, requires continuous monitoring, and introduces the risk of leakage between the pleural cavity and the water seal, which is a fatal complication. [11] In addition, it does not decrease the time of medical management. [5,8,11] Lavage with 10 to 25 mL/kg sterile saline warmed to 38°C has been recommended in recently published literature, [4,5,11,22] although there are no controlled prospective studies examining the outcome. A study

published in 2002 recommended against lavage due to the risk of a nosocomial antimicrobial-resistant infection as well as the risk of being unable to recover fluid instilled. [8] In theory, lavage maintains patency of the tubes, facilitates drainage and debridement of the pleura, and dilutes bacteria and inflammatory mediators. [11,14,22] One study reported that using a lavage solution for treatment of pyothorax resulted in earlier tube removal, but increased frequency of lavage did not correlate with shorter tube duration. [10] There are no definitive guidelines for the frequency of lavage – every four hours for the first 24 to 48 hours and then two to three times daily has been recommended. [4,11,22] The addition of fibrinolytics or antimicrobials to the lavage solution is not recommended based on outcomes in human pyothorax. [1,11,22]

Pending culture and sensitivity results, antimicrobials selected should be effective against both obligate and facultative anaerobes. [9,11,19] Consideration should also be given to choosing antimicrobials with low toxicity and good distribution to the pleural space. [9] Good initial antimicrobial choices include penicillin G, ampicillin, amoxicillin, ticarcillin-clavulanic acid, ampicillin-sulbactam, or amoxicillin-clavulanic acid, as most anaerobic isolates are susceptible to these. [9,11,19] Monotherapy is generally effective, and there is no need to treat gram-negative facultative bacteria with an additional antibiotic such as a quinolone or an aminoglycoside as enteric organisms are rarely isolated from feline pleural fluid samples. [11] Most gram-negative rods identified on cytologic analysis are *Pasteurella* spp, which are susceptible to penicillin and its derivatives. [5,9,11,22] In addition, aminoglycosides are nephrotoxic, while enrofloxacin is potentially retinotoxic. [5] Poor choices for empiric monotherapy include first generation cephalosporins and clindamycin; while they are effective against anaerobes, they have poor efficacy against *Pasteurella* spp. [11,22] Antibiotics should ideally be given initially via an intravenous route, and oral antibiotics can be utilized when the patient is clinically improved and eating well. [4,11,14] High doses of antimicrobials are recommended for treating anaerobic infections, as they are associated with devitalized tissue with poor perfusion. [14,22] In addition, the duration of antibiotic therapy is usually prolonged, as cats with pyothorax are prone to relapse if antibiotic therapy is discontinued prematurely. [5,10,22] Antimicrobial therapy should be administered for four to six weeks, and thoracic radiographs should be taken prior to discontinuing antibiotics to verify complete resolution of pyothorax. [5,10,11,22]

Opioid analgesia should be provided prior to anesthetic induction and after drain placement. [11,22,24] Opioids are good choices in critically ill patients because they have rapid onset and are safe, reversible, and potent analgesics. [24] Non-steroidal anti-inflammatory drugs are excellent for reducing pain associated with peripheral inflammation, as they inhibit cyclooxygenase enzyme isoforms (COX). COX-1 is responsible for basal prostaglandin production for normal homeostatic processes within the body, while COX-2 is found at sites of inflammation. [24] There are no pure COX-2 inhibitors currently available, however. As a result, nonsteroidal anti-inflammatory drugs may not be appropriate as a source of pain management for cats with pyothorax at initial presentation, as they are often hypotensive and hypovolemic. [24] Intrapleural analgesia is not recommended due to the potential for diaphragmatic paralysis in patients with compromised respiratory reserve, as well as unreliable absorption due to the presence of an inflammatory effusion. [11,22,24]

Close monitoring of patients with indwelling thoracostomy tubes is recommended for complications such as pneumothorax, failure of drainage, subcutaneous edema, abscesses at the site of drain insertion, or movement of drains. [11] In addition, daily monitoring should include measurement of electrolytes, hematocrit, serum albumin, total plasma protein and body weight. [11] Tubes typically remain in place a median of five to six days. [5,8,10] Criteria for tube removal include minimal residual effusion on aspiration, resolution of pleural effusion on thoracic radiographs, and resolution of infection based on cytology (absence of microorganisms, reduction in neutrophil numbers and a decrease in toxic or degenerate appearance). [11,14]

Prognosis for cats treated for pyothorax is good if they survive the first 48 hours after initial presentation, with the vast majority of fatalities occurring during this period. [5,8,10] In two studies, 100% of deaths occurred within the first 48 hours [5,10]; in a second study, 66% of deaths occurred within the first 24 hours. [8] Poor prognostic indicators include hypersalivation, bradycardia, and hypothermia combined with bradycardia. [5] Overall survival rates for cats treated for pyothorax range from 66% to 78%. [5,8] Success rate following placement of closed thoracostomy tubes is high, ranging from 73% to 95%. [5,8,13] Recurrence of pyothorax is very rare, and additional treatment (medical or surgical) is usually curative. [8,10,11,25]

Thoracotomy is rarely required to treat feline pyothorax due to the high success rate of medical management using closed tube thoracostomy. Indications for surgery include pulmonary or mediastinal abscessation, extensively loculated effusions, marked lung lobe consolidation, persistence of large-volume effusions after more than seven days, drain obstruction, or recurrence of pyothorax after withdrawal of antibiotics. [11,12,25] Thoracotomy, when necessary, is associated with a high success rate, with studies reporting 100% successful outcomes. [5,8,12,25] Right- or left-sided pneumonectomy in cats is well tolerated. [5,8,25] Advantages of surgery over medical management include full surgical exploration of the thoracic cavity, removal of all exudates from the pleural space with lavage and debridement, identification and removal of any foreign material, and correct placement of indwelling thoracostomy tubes. [1,5,8] Disadvantages of surgery include increased cost compared to medical management, pain associated with the surgery, and risk of prolonged general anesthesia in septic cats. [8]

Clinical Report

An eight-year-old neutered male domestic shorthair cat presented for lethargy and inappetence of two days duration. His owner reported that a similar episode had occurred approximately six weeks previously that appeared to resolve after two days. He had no history of coughing, sneezing, vomiting or diarrhea, but his water intake was reported by his owner to be decreased. He was an indoor-only cat, and his owner could not recall his vaccination history. He was the only cat in the household. On presentation, the cat was quiet and mildly febrile at 39.0° C, with a respiratory rate of 44 breaths per minute and a heart rate of 160 beats per minute. He weighed 5.8 kg with a body condition score of five out of nine. The cat had a restrictive breathing pattern and there were diminished breath sounds ventrally on auscultation. In addition, he had decreased skin turgor and slightly tacky mucous membranes. The abdomen was normal on palpation.

Based on the history and physical examination, pleural effusion was suspected and two view thoracic radiographs were obtained (Figure 1a and 1b). These showed a large volume of pleural effusion on the right side of the thorax, obscuring the cardiac silhouette. A fluid line was visible on the lateral view, with an opacified area present in the caudodorsal lung lobe, and the lung lobes were retracted from the thoracic wall. The visible cardiac silhouette and pulmonary vasculature appeared normal, and no mass lesions were noted. Differential diagnoses for the pleural effusion included hemorrhage (trauma, coagulopathy), exudate (infection), transudate (hypoproteinemia), and modified transudate (neoplasia, congestive heart failure, fungal infection, feline infectious peritonitis, systemic inflammation, chylothorax).

Figure 1a – Thoracic Radiographs Prior to Thoracostomy Tube Placement

Right Lateral

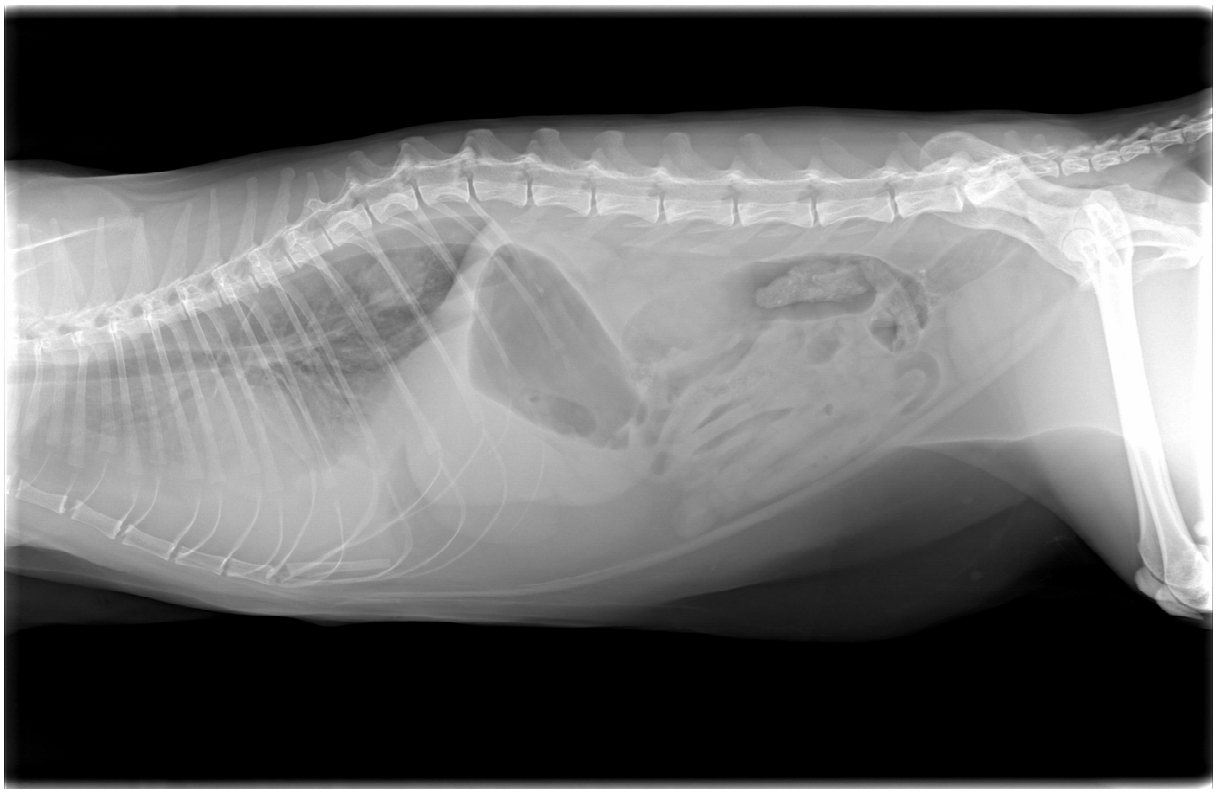
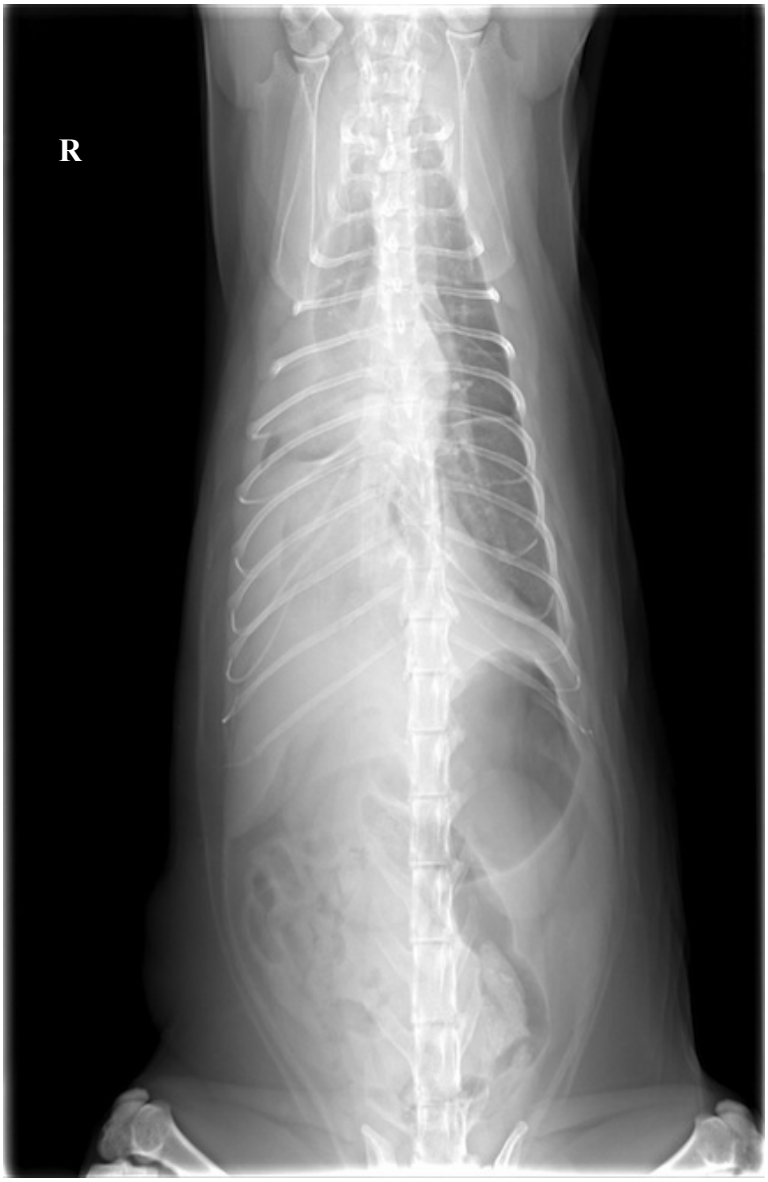


Figure 1b – Thoracic Radiographs Prior to Thoracostomy Tube Placement

Dorsoventral



Thoracic ultrasound was performed with the cat in sternal recumbency. [(a)] A small amount free fluid was identified on the left side of the chest, while a larger volume of fluid and a consolidated lung lobe were visible on the right side of the chest. No mass lesions, cardiomegaly or left atrial enlargement were noted. The cat's right chest wall was shaved and aseptically prepared in an approximate seven cm band between the fifth and tenth ribs. Thoracocentesis was performed using a 19-gauge butterfly needle on the right side of the chest between the seventh and eighth rib space; only three to four mL of fluid could be aspirated due to the highly viscous nature of the effusion. The fluid aspirated was pink, thick and foul smelling. Samples were placed in a lavender-top (EDTA) tube for cytology and fluid analysis, in a red top tube and on two sterile culturettes for aerobic and anaerobic culture and sensitivity, and on a glass slide for in-house cytology. The slide was stained using modified Wright-Giemsa. In-house cytologic evaluation showed sheets of degenerate neutrophils with intracellular and extracellular bacteria and reactive mesothelial cells. Based on thoracic radiographs, thoracic ultrasound, gross fluid appearance and in-house cytology, a diagnosis of pyothorax was made. Thoracic fluid analysis, cytology, and aerobic and anaerobic cultures were submitted to an outside laboratory for analysis. [(b)]

In-house serum chemistries [(c)] (Table 1) showed mildly decreased alkaline phosphatase (12 U/L, 14-111 reference range), and an elevated glucose (166 mg/dL, 74-159 reference range). The low alkaline phosphatase was considered likely due to artifact, while the mild elevation in glucose was attributed to stress hyperglycemia.

An in-house complete blood count (Table 2)[(d)] showed leukocytosis (total white blood cell count of 51.6 K/μL, 5.5 – 19.5 reference range), neutrophilia (47.07 K/μL, 2.5 – 12.5 reference range), monocytosis (3.00 K/μL, 0.15 – 1.7 reference range), and thrombocytopenia (151 K/μL, 175 – 600 reference range). The primary differential diagnoses for severe leukocytosis included infection (pyothorax) and severe inflammation (necrosis). Differential diagnoses for thrombocytopenia included consumption (disseminated intravascular coagulation) and artifact (platelet clumping). An in-house combination feline leukemia virus antigen and feline immunodeficiency virus antibody test (Table 2) [(e)] was negative. An in-house electrolyte test (Table 2) [(f)] was performed, and sodium was slightly lower than the reference range (143 mmol/L, reference range 147 – 162 mmol/L), while potassium and chloride were within the reference ranges. Differential diagnoses for hyponatremia included hypovolemia and loss of fluid into the thoracic cavity.

Table 1 – In House Blood Test Results Day One

| Test | Laboratory Value | Reference Range |
|---|---|---|
| *Serum Chemistries:* | | |
| Albumin | 3.3 g/dL | 2.2 - 4 |
| **Alkaline phosphatase** | **12 U/L (L)** | 14 - 111 |
| Alanine aminotransferase | 31 U/L | 12-130 |
| Amylase | 716 U/L | 500 – 1,500 |
| Blood urea nitrogen | 25 mg/dL | 16 - 36 |
| Calcium | 9.3 mg/dL | 7.8 – 11.3 |
| Creatinine | 1.3 mg/dL | 0.8 – 2.4 |
| **Glucose** | **166 mg/dL (H)** | 74 - 159 |
| Lipase | 1,356 U/L | 100 – 1,400 |
| Phosphorous | 5.6 mg/dL | 3.1 – 7.5 |
| Total bilirubin | 0.3 mg/dL | 0 – 0.9 |
| Total protein | 7.4 g/dL | 5.7 – 8.9 |
| Globulins | 4.1 g/dL | 2.8 – 5.1 |

Table 2 – In House Blood Test Results Day One

| Test | Laboratory Value | Reference Range |
|---|---|---|
| *Complete Blood Count:* | | |
| **WBC** | **51.60 K/µL (H)** | 5.5 – 19.5 |
| RBC | 6.08 M/µL | 5 - 10 |
| HGB | 12.8 g/dL | 9 – 15.1 |
| HCT | 32.2% | 30 – 45% |
| MCV | 46.1 fL | 41 - 58 |
| MCH | 18.30 pg | 12 - 20 |
| **Absolute Neutrophil Seg** | **47.07 K/µL (H)** | 2.5 – 12.5 |
| Absolute Lymphocytes | 0.72 k/µL | 0.4 – 6.8 |
| **Absolute Monocytes** | **3.00 K/µL (H)** | 0.15 – 1.7 |
| Absolute Eosinophils | 0.71 K/µL | 0.1 – 0.79 |
| Absolute Basophils | 0.1 K/µL | 0 – 0.1 |
| **Platelets** | **151 K/µL (L)** | 175 – 600 |
| | | |
| *Retroviral Testing:* | | |
| FeLV Antigen | Negative | Negative |
| FIV Antibody | Negative | Negative |
| | | |
| *Electrolytes:* | | |
| **Sodium** | **143 mmol/L (L)** | 147 - 162 |
| Potassium | 3.9 mmol/L | 2.9 – 4.2 |
| Chloride | 115 mmol/L | 112 - 129 |

A 22-guage intravenous catheter was placed in the right cephalic vein and the cat was placed on an isotonic crystalloid replacement solution [(g)] at a rate of two mL/kg/hour intravenously (IV) for approximately two hours prior to anesthetic induction for thoracostomy tube placement. Due to anorexia and hypovolemia, it was believed that the cat was likely whole-body potassium depleted, and 20 mEq/L potassium chloride was added to the fluids. Ticarcillin-clavulanic acid [(h)] was commenced (50 mg/kg IV q6h), and a pre-anesthetic pain medication was administered (buprenorphine HCl, 0.02 mg/kg IV). Prior to induction of anesthesia, systolic blood pressure was measured from the left radial artery at 75 mmHg using ultrasonic Doppler [(i)] and a three mm cuff. The cat received a 20 mL/kg bolus of an isotonic replacement crystalloid solution [(g)] over 15 minutes prior to anesthetic induction, and its systolic blood pressure rose to 90 mmHg. The cat was pre-oxygenated for five minutes with 100% flow-by oxygen, general anesthesia was induced with propofol [(j)] (four mg/kg IV), the cat was intubated, and anesthesia was maintained using inhalant isoflurane. Isotonic crystalloids [(g)] were administered at a rate of 10 mL/kg/hour IV to support the cat's blood pressure during the procedure. The cat's blood pressure, heart rate and oxygen saturation were monitored every five minutes during the procedure. [(k)] The cat's right chest wall was shaved and aseptically prepared, a local anesthetic block of the skin and subcutaneous tissue was performed with 0.9 mg/kg of 2% lidocaine at the $12^{th}$ rib, and the intercostal muscles were blocked with a similar amount of lidocaine between the $10^{th}$ and $11^{th}$ intercostal spaces. Standard sterile technique was utilized during the procedure. A small skin incision was made mid-thorax over the 12th intercostal space. An 8 French thoracostomy tube with a stylette [(l)] entered the skin at the $12^{th}$ intercostal space and was advanced subcutaneously to between the $10^{th}$ and $11^{th}$ rib space, where it entered the thoracic cavity. The thoracostomy tube was advanced off the stylette and fed cranially, and the stylette was removed. The tube was clamped shut with a c-clamp, and a Christmas tree connector and three-way stop-cock were placed on the end of the thoracostomy tube. These were sutured together with 2-0 nylon to prevent inadvertent dislodgement. The tube was secured to the cat's skin using a purse string suture and a Chinese finger trap utilizing 2-0 nylon suture material. A second thoracostomy tube was placed in the cat's left thorax using an identical procedure. Warm 0.9% sterile saline was flushed in and out of the thoracostomy tubes until the fluid returning appeared mostly clear. A non-adherent pad coated in povidone-iodine ointment was placed against each thoracostomy tube, and a surgical stockinet was fitted from the neck to the abdomen.

During the procedure, the cat's systolic blood pressure dropped to 70 mmHg, and two slow five mL/kg IV boluses of a colloid solution [(m)] were administered. Three-view thoracic radiographs were repeated after thoracostomy tube placement to confirm positioning of the thoracostomy tubes (Figure 2a, 2b and 2c). The left tube was not positioned as cranially as desired, and the right tube appeared to cross the mediastinum to the left side, but flushing was highly productive. Because the patient had become hypotensive under anesthesia, repositioning of the thoracostomy tubes was not pursued.

Figure 2a – Thoracic Radiographs After Thoracostomy Tube Placement

Left Lateral

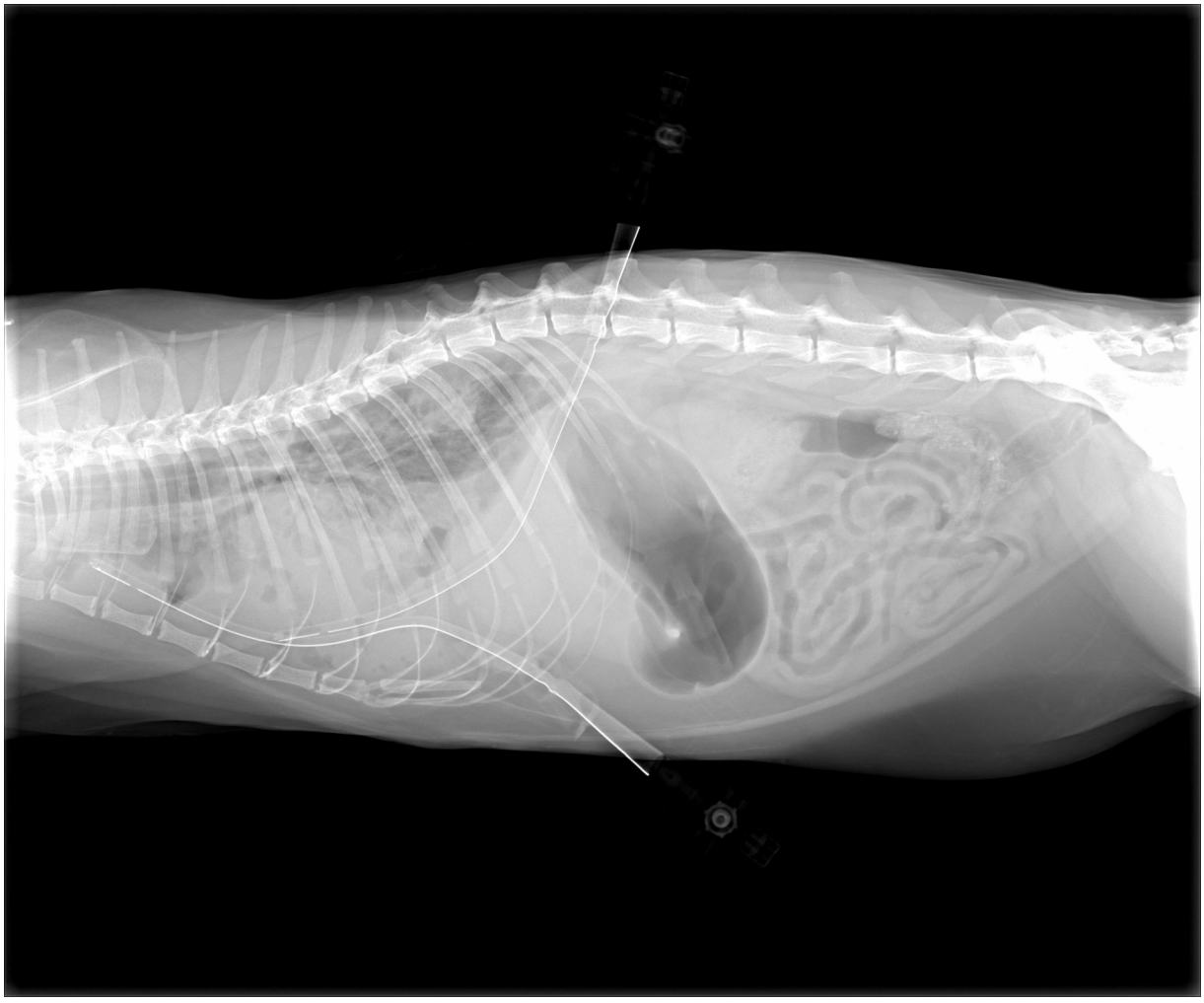


Figure 2b – Thoracic Radiographs After Thoracostomy Tube Placement

Right Lateral

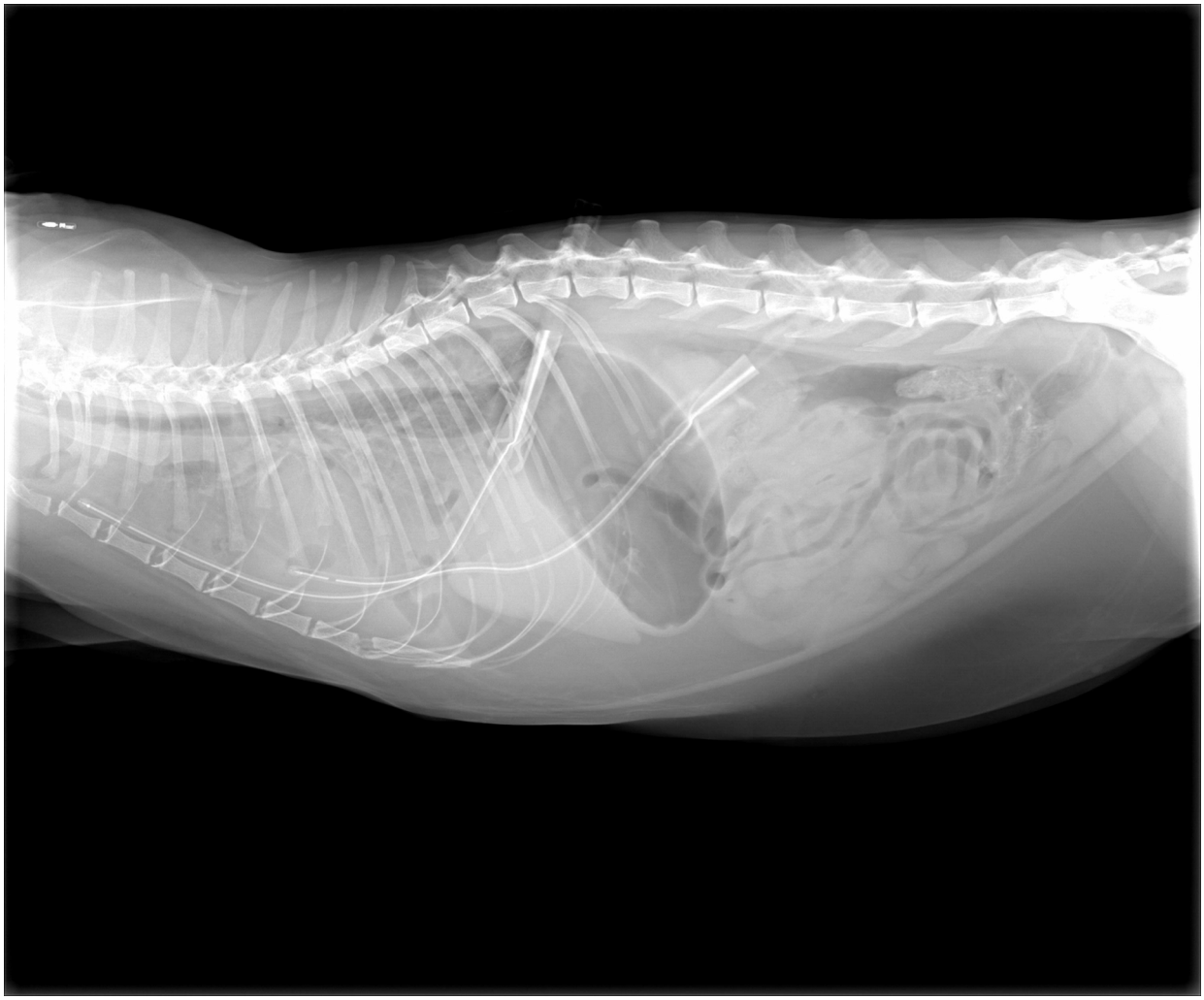
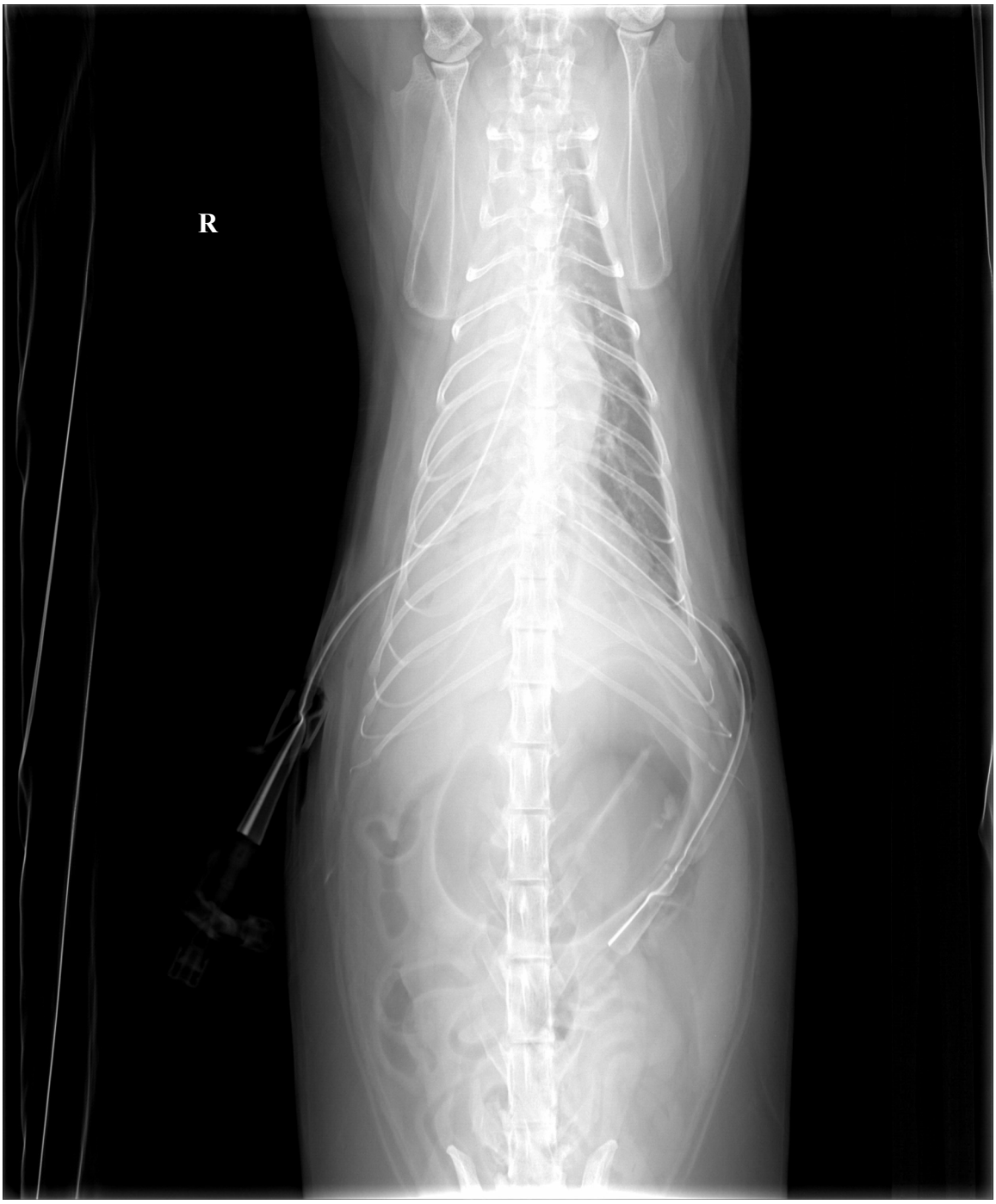


Figure 2c – Thoracic Radiographs After Thoracostomy Tube Placement

Dorsoventral



The cat's systolic blood pressure dropped to 50 mmHg after extubation and a dopamine HCl continuous rate infusion was started at five mcg/kg/minute; the dopamine was increased in one mcg/kg/minute increments every five minutes until the cat's systolic blood pressure stabilized at 75 mmHg while on 10 mcg/kg/minute. The cat recovered from anesthesia in a warm oxygen cage [(n)] with 40% inhaled oxygen.

Post-operatively, the cat was administered intravenous isotonic crystalloid replacement fluids [(g)] supplemented with 20 mEq/L potassium chloride at a rate of two mL/kg/hour. The patient was administered buprenorphine HCl at 0.02 mg/kg subcutaneously (SQ) q6h for pain management. Thoracostomy tube care consisted of flushing a total of 17 mL/kg warm 0.9% sterile saline into each thoracostomy tube in 25 mL aliquots every four hours, with aspiration performed after each flush. Flush recovered from the right thoracostomy tube appeared repeatedly bloodier with larger chunks of material compared to the tube in the left side. Ticarcillin-clavulanic acid [(h)] was administered (50 mg/kg IV q6h) and respiratory rate and effort were monitored every two hours. Dopamine was administered in a constant rate infusion at 10 mcg/kg/minute IV. Blood pressure was not measured overnight.

The following day (day two), the cat was afebrile (38.0° C), bright and alert. His respiratory rate and effort were mildly elevated (30 breaths per minute), his heart rate was 200 beats per minute, and he reacted painfully when touched near his thoracostomy tubes. His physical examination revealed mild generalized subcutaneous edema, although his weight was stable at 5.77 kg. Differentials for generalized subcutaneous edema included fluid overload, vasculitis, and hypoalbuminemia.

An in-house electrolyte test [(f)] in the morning showed on-going mild hyponatremia (144 mmol/L, 147-162 reference range) and low normal potassium (3.0 mmol/L, 2.9 – 4.2 reference range) (Table 3). Differential diagnoses for low normal potassium despite IV supplementation included decreased dietary potassium intake, potassium loss into the thoracic cavity, and dilution due to intravenous fluid therapy. Packed cell volume was within normal limits, but total solids were slightly low (5.8 g/dL; 5.9 – 8.5 reference range). Differentials for hypoproteinemia included dilutional effects of IV fluid therapy, loss of albumin into the pleural space, and decreased nutritional intake.

Table 3– In-house Blood Work Day Two 8:00 a.m.

| Test | Laboratory Value | Reference Range |
|---|---|---|
| *Electrolytes:* | | |
| **Sodium** | **144 mmol/L (L)** | 147 – 162 |
| Potassium | 3.0 mmol/L | 2.9 – 4.2 |
| Chloride | 123 mmol/L | 112 - 129 |
| *PCV/TS:* | | |
| Packed Cell Volume % | 33% | 29 – 45% |
| **Total Solids** | **5.8 g/dL (L)** | 5.9 – 8.5 g/dL |

Potassium chloride supplementation was increased from 20 to 30 mEq/L; the intravenous fluid rate was unchanged at two mL/kg/hour. The cat's morning systolic blood pressure was measured at 100 mmHg, and the dopamine continuous rate infusion was decreased to eight mcg/kg/minute. The cat's blood pressure was rechecked hourly and if the blood pressure was greater than 90 mmHg, the dopamine rate was decreased an additional two mcg/kg/minute. The dopamine was discontinued after five hours, and the cat's blood pressure was then monitored every two hours. The blood pressure remained between 90 and 105 mmHg, and blood pressure monitoring was discontinued at 6 p.m.

During day two, the thoracostomy tube insertion sites were inspected for inflammation and to ensure that the sutures remained in place; the insertion sites were cleaned with a chlorhexidine solution and a fresh non-adherent pad was replaced at each location. No changes were made to the thoracostomy tube flushing protocol or to antibiotic therapy. The flushed exudate from the right thoracostomy tube remained bloody with thick chunks; the left thoracostomy tube flush became lighter pink with less visible material present. Because the thoracostomy tubes appeared to be causing the patient significant discomfort, buprenorphine HCl was discontinued and a continuous rate infusion of fentanyl citrate was initiated with a three mcg/kg bolus IV and maintained with a continuous rate infusion at three mcg/kg/hour. The cat did not show interest in food on day two.

Electrolytes were rechecked in-house [(f)] (Table 4) at the end of day two, and they showed normalization of potassium (4.0 mmol/L, 2.9 – 4.2 reference range) and improvement in sodium (145 mmol/L, 147 – 162 reference range). Total solids were lower (5.0 g/dL, 5.9 – 8.5 reference range), likely due to the on-going dilutional effects of IV fluid therapy and loss of albumin into the pleural space.

Thoracic fluid cytology and fluid analysis results returned from the reference laboratory [(b)] at the end of day two, which showed a severe suppurative exudate with intra- and extra-cellular rod bacteria (Table 5), confirming the diagnosis of pyothorax. Antibiotic therapy was not changed from ticarcillin-clavulanic acid [(h)] based on these results, as the most likely bacteria were thought to be sensitive to it.

Table 4 – In-house Blood Work Day Two 6:30 p.m.

| Test | Laboratory Value | Reference Range |
|---|---|---|
| *Electrolytes:* | | |
| **Sodium** | **145 mmol/L (L)** | 147 – 162 |
| Potassium | 4.0 mmoL | 2.9 – 4.2 |
| Chloride | 120 mmol/L | 112 - 129 |
| *PCV/TS:* | | |
| Packed Cell Volume % | 34% | 29 – 45% |
| **Total Solids** | **5.0 g/dL (L)** | 5.9 – 8.5 g/dL |

Table 5 – Thoracic Fluid Analysis Day Two

| Test | Result |
|---|---|
| Protein | 4.5 g/dL |
| RBC | < 100,000 cells/uL |
| Volume | 8.0 mL |
| WBC | 465,790 cells/uL |
| Color | Pale brown/opaque |
| Interpretation | Exudate |

*Description: Marked septic suppurative inflammation. There is a marked increase in inflammatory cells that are primarily variably degenerate neutrophils with a low number of macrophages. There are many mixed bacteria found scattered in the background and in occasional thick clumps in the background. They are also found within neutrophils. The majority are mixed rods, some that form chains. Some are slender rods. No other organisms are identified and no foreign material or neoplastic cells are observed. These findings indicate marked inflammation and bacterial infection.*

On the third day of hospitalization, the cat was afebrile (38.0° C), had a mildly elevated respiratory rate (32 breaths per minute) but normal effort. He weighed 5.78 kg and continued to have generalized subcutaneous edema. Electrolytes (sodium, potassium and chloride) were rechecked in-house [(f)] in the morning (Table 6), and all electrolytes were within the reference range (sodium 147 mmol/L, 147 – 162 reference range; potassium 3.4 mmol/L, 2.9 – 4.2 reference range; chloride 124 mmol/L, 112 – 129 reference range). Total solids, while still decreased, had improved (5.8 g/dL, 5.9 – 8.5 reference range).

The cat's on-going subcutaneous edema was attributed to mild fluid overload, so his IV fluids were discontinued and his IV catheter was flushed with heparanized saline q6h. Because the cat appeared to be experiencing less discomfort associated with his thoracostomy tubes, his pain protocol was changed from an intravenous continuous rate infusion of fentanyl citrate to buprenorphine HCl 0.02 mg/kg SQ q6h. Meloxicam [(o)] was administered at a dose of 0.1 mg/kg orally (PO) one time. Thoracostomy tube lavage was decreased from every four hours to every six hours (flushing 17 mL/kg of warm sterile saline in 25 mL aliquots and aspirating back after each flush); the aspirated flush from both the left and right thoracostomy tubes appeared less cloudy and sanguineous than on day two. The thoracostomy tube insertion sites were inspected and cleaned once daily. He continued to receive ticarcillin-clavulanic acid [(h)] at 50 mg/kg IV q6h. He began to eat small amounts of canned food and to drink during the day.

Fluid was collected from the right thoracostomy tube for in-house cytologic evaluation. It was placed on a slide and stained with a modified Wright-Giemsa stain, and showed many neutrophils with occasional intracellular bacteria. More macrophages were present compared to previous in-house evaluation, occasionally with intracellular neutrophils.

Repeat two-view thoracic radiographs obtained on day three showed moderate improvement in the opacity in the right thorax, gas distension of the stomach and small intestines, and a more cranial positioning of the left thoracostomy tube (Figure 3a and 3b).

The culture and sensitivity at the reference laboratory [(b)] reported no growth.

Table 6 – In-house Blood Work Day Three 8:00 a.m.

| Test | Laboratory Value | Reference Range |
|---|---|---|
| *Electrolytes:* | | |
| Sodium | 147 mmol/L | 147 - 162 |
| Potassium | 3.4 mmol/L | 2.9 – 4.2 |
| Chloride | 124 mmol/L | 112-129 |
| *PCV/TS:* | | |
| Packed Cell Volume % | 34% | 29 – 45% |
| **Total Solids** | **5.8 g/dL (L)** | 5.9 – 8.5 g/dL |

Figure 3a – Thoracic Radiographs Day Three

Right Lateral

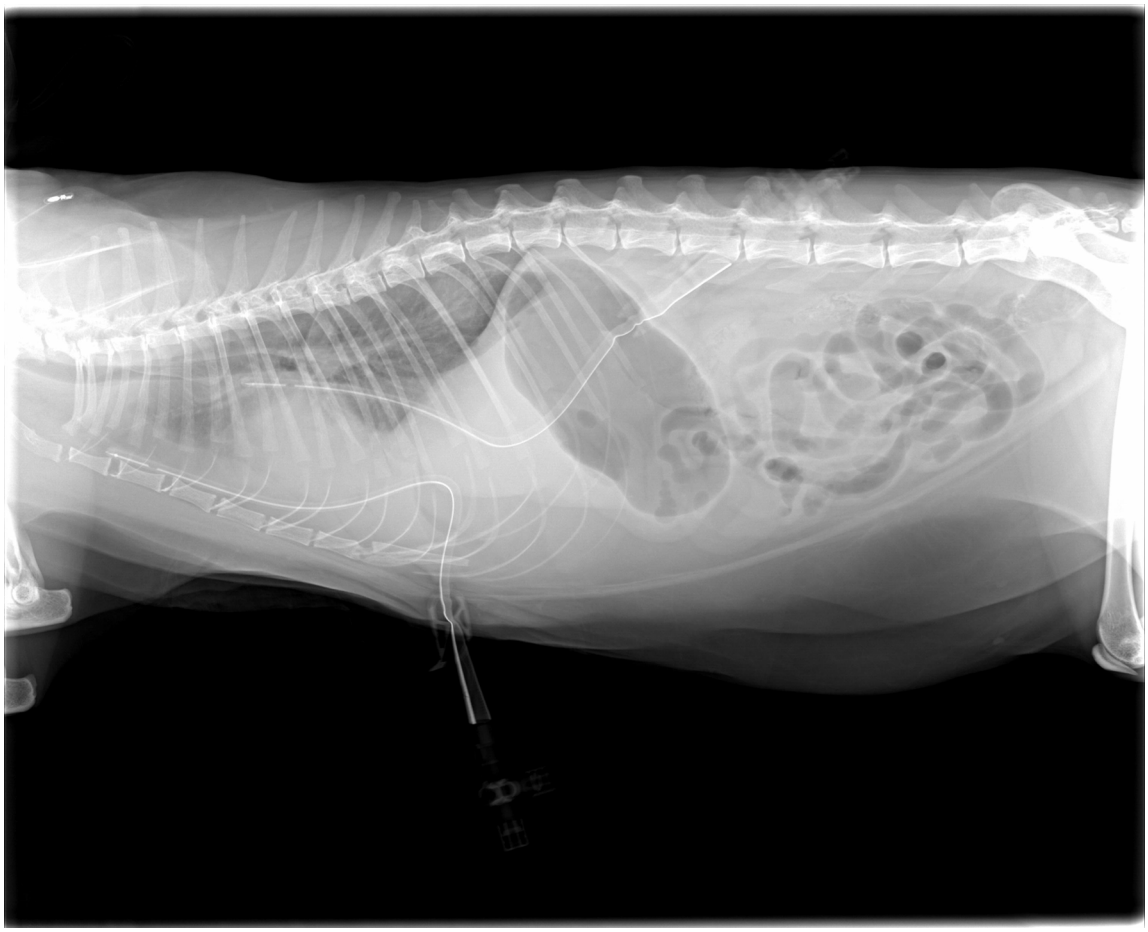
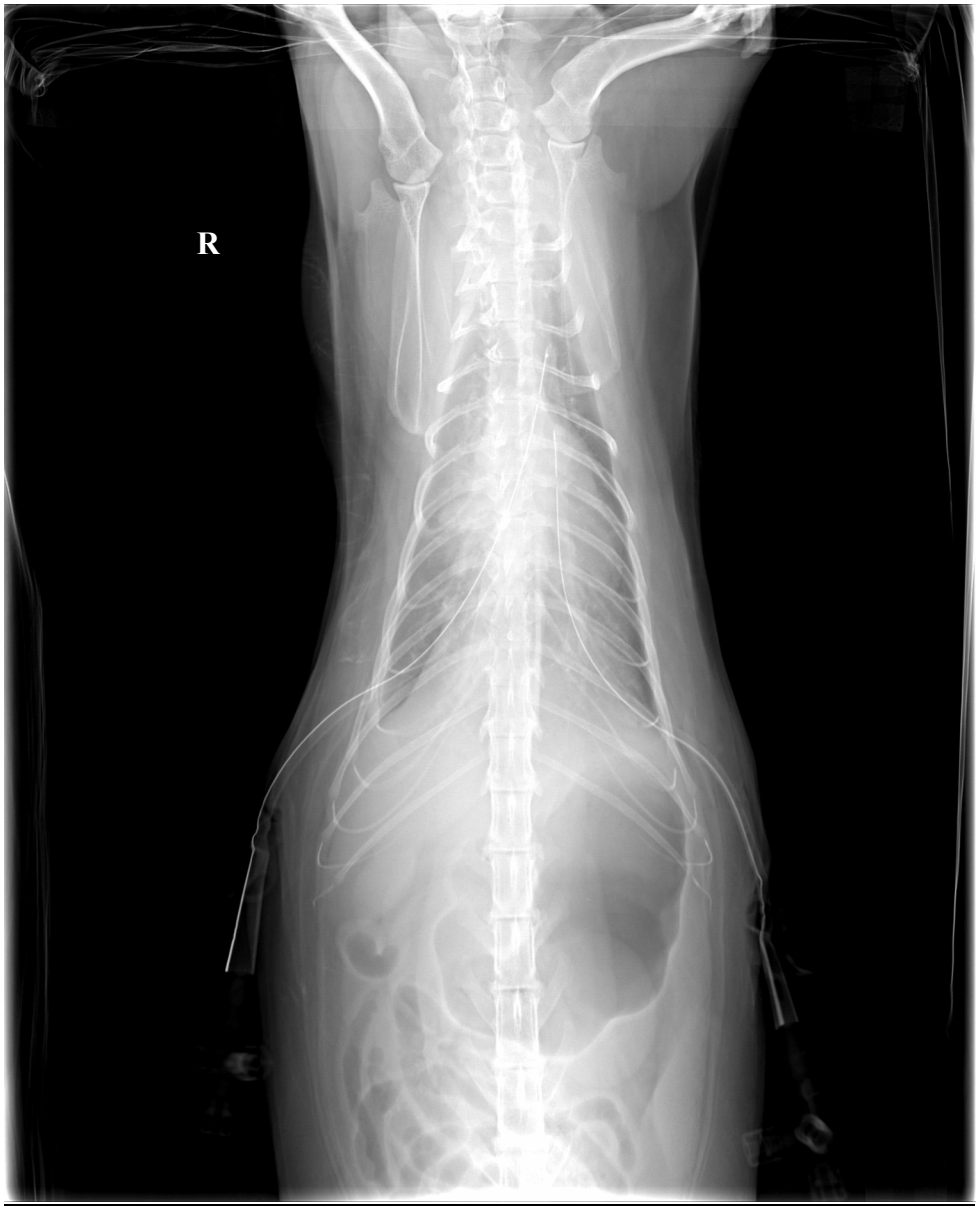


Figure 3b – Thoracic Radiographs Day Three

Ventrodorsal



On day four of hospitalization, the cat was afebrile (38.0° C), had a mildly elevated respiratory rate (34 breaths per minute) with normal effort. He weighed 5.79 kg. He still had mild generalized subcutaneous edema. He continued to eat and drink. His IV catheter was removed and his antibiotics were changed from IV ticarcillin-clavulanic acid [(h)] to oral amoxicillin-clavulanic acid [(p)] (16 mg/kg PO q12h). Thoracostomy tube lavage was reduced from every six hours to every eight hours with no other changes in the daily protocol. The aspirated material continued to have some flocculent material present, but did not appear grossly sanguineous. The pain management protocol was changed from subcutaneous administration of buprenorphine HCl to oral administration (0.02 mg/kg to buccal mucosa q8h). Electrolytes (sodium, potassium and chloride), packed cell volume and total solids were rechecked in-house, [(f)] and all values were within the reference range (Table 7).

A recheck complete blood count was submitted to the reference laboratory [(b)] on day four (Table 8), which showed a mature neutrophilia with toxic changes (27,360 neutrophils/μL, 2,500-12,500 reference range), a left shift (5,320 bands/μL, 0-300 reference range), and a monocytosis (2,280 monocytes/μL, 0-850 reference range). The total white blood cell count was significantly improved (38.0 k/μL total white blood cells, 4.2 – 15.6 reference range) compared to that obtained at hospital admission (51.6 k/μL total white blood cells). The primary differential for the leukocytosis was on-going infection of the pleural space with secondary inflammation.

No growth was reported on the aerobic and anaerobic culture and sensitivity on day four from the reference laboratory. [(b)]

Table 7 – In-house Blood Work Day Four 8:00 a.m

| Test | Laboratory Value | Reference Range |
|---|---|---|
| *Electrolytes:* | | |
| Sodium | 155 mmol/L | 147 - 162 |
| Potassium | 4.0 mmol/L | 2.9 – 4.2 |
| Chloride | 126 mmol/L | 112-129 |
| *PCV/TS:* | | |
| Packed Cell Volume % | 34% | 29 – 45% |
| Total Solids | 6.4 | 5.9 – 8.5 g/dL |

Table 8: Complete Blood Count on Day Four

| Test | Laboratory Value | Reference Range |
|---|---|---|
| **WBC** | **38.0 K/µL (H)** | 4.2 – 15.6 |
| RBC | 6.87 M/µL | 6.0 – 10.0 |
| HGB | 10.8 g/dL | 9.5 - 15 |
| HCT | 34.3% | 29 - 45 |
| MCV | 50 fL | 41 - 58 |
| MCH | 15.7 pg | 11.0 – 17.5 |
| MCHC | 31.5 g/dL | 29 - 36 |
| Neutrophil Seg | 72% | 35 - 75 |
| **Neutrophil Bands** | **14% (H)** | 0 - 3 |
| **Lymphocytes** | **8% (L)** | 20 - 55 |
| **Monocytes** | **6% (H)** | 1 - 4 |
| Eosinophils | 0% | 2 - 12 |
| Basophils | 0% | 0 - 1 |
| Platelets | 209 K/µL | 170 - 600 |
| **Absolute Neutrophils** | **27,360 /µL (H)** | 2,500 – 12,500 |
| **Absolute Neutrophil Bands** | **5,320/µL (H)** | 0 - 300 |
| Absolute Lymphocytes | 3,040 µL | 1,500 – 7,000 |
| **Absolute Monocytes** | **2,280 /µL (H)** | 0 - 850 |
| Absolute Basophils | 0 µL | 0 - 100 |

*Remarks: slide reviewed microscopically – neutrophils appear slightly toxic*

On the fifth and sixth day of hospitalization, the cat was afebrile with mildly elevated respiratory rate and normal respiratory effort. No laboratory work was performed, as the pet was eating small amounts and drinking, although his weight was declining (5.7 kg on day five, 5.62 kg on day six). He developed soft stools on day five, which were attributed to either dietary change in the hospital or to amoxicillin-clavulanic acid. (p) Antibiotic therapy, pain management protocols and thoracostomy tube maintenance was unchanged. Flocculent material was still noted in the fluid aspirated back from both the right and left thoracostomy tubes. Mirtazapine was added to his therapy on day six (0.66 mg/kg PO q72h) in an attempt to improve his appetite, which appeared to result in increased nutritional intake.

Aerobic and anaerobic culture and sensitivity were available from the reference laboratory (b) on day five, which showed isolation of a single aerobic organism (*Actinomyces* spp) and a single anaerobic organism (*Fusobacterium* spp) (Table 9). Amoxicillin-clavulanic acid (p) was continued as antibiotic monotherapy based on the results.

Table 9 – Microbiology Results Day Five

| | |
|---|---|
| Aerobic Results: | |
| Actinomyces species 1+ | Successful treatment requires prolonged antibiotic administration. Antimicrobial of choice: Penicillin (high doses), Erythromycin, Clindamycin, Ampicillin, Doxycycline and Cephalosporins are effective. (The culture characteristics of this organism prohibits standardized susceptibility testing). |
| Anaerobic Results: | |
| Fusobacterium species 2+ | Antimicrobial of choice: Amoxicillin or Clavamox ($1^{st}$), Clindamycin, Metronidazole or Chloramphenicol ($2^{nd}$) |

On day seven, the cat's temperature was 38.4° C, and he appeared bright and alert. His respiratory rate was 34 breaths per minute. He weighed 5.67 kg. Aspirate from the thoracostomy tube showed minimal flocculent material, and the decision was made to remove the thoracostomy tubes. The cat was sedated with 1.2 mg/kg teletamine HCl [(q)] intramuscularly. The sutures holding the thoracostomy tube in places were cut and the thoracostomy tubes were removed. The insertion sites at the skin were closed with surgical staples and the cat was recovered in an oxygen cage [(n)] for two hours where his respiratory rate and effort were closely watched. He recovered uneventfully and was discharged to the owner later in the afternoon. He was discharged on a six-week course of amoxicillin-clavulanic acid [(p)] (16.4 mg/kg PO q12h). Analgesia (buprenorphine) was discontinued after day seven.

The cat presented for a recheck examination one week after discharge. His owner reported that he was eating well, showed daily improvements in his energy level, and was breathing normally at home. He continued to have soft stools but was not experiencing any vomiting. On physical examination, he was afebrile (38.3° C), had a mildly increased respiratory rate (40 breaths per minute) with normal effort, and ausculted slightly more dully on the right thorax compared to the left. His weight had decreased to 5.56 kg. The surgical staples were still in place and the thoracostomy tube skin incisions appeared healed. Repeat thoracic radiographs showed resolution of his pyothorax (Figure 4a and 4b).

Figure 4a – Thoracic Radiographs Seven Days After Thoracostomy Tube Removal

Right Lateral

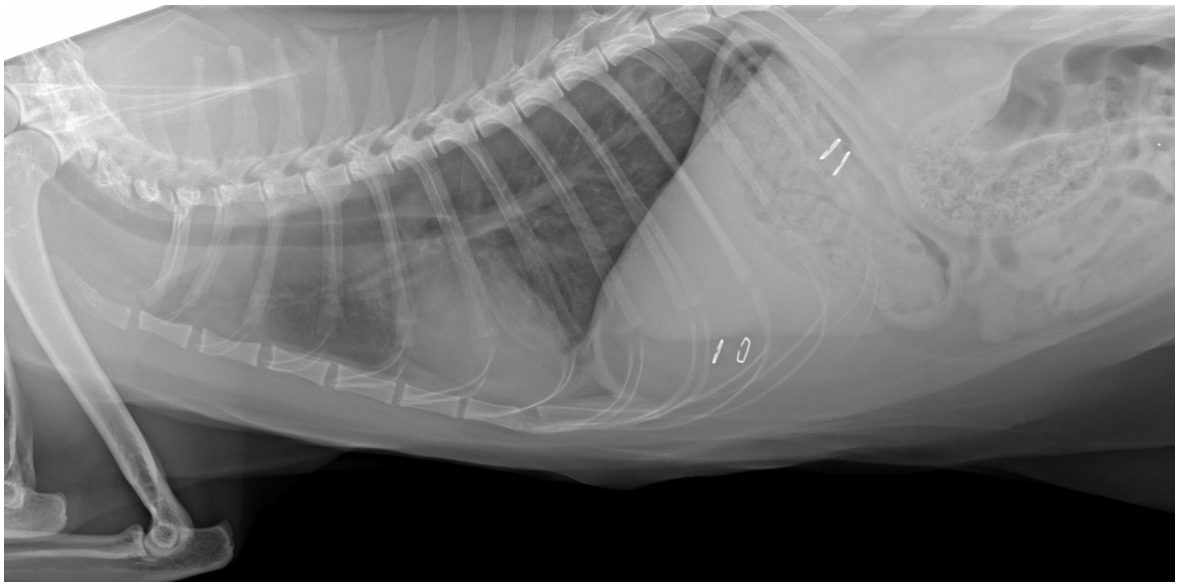
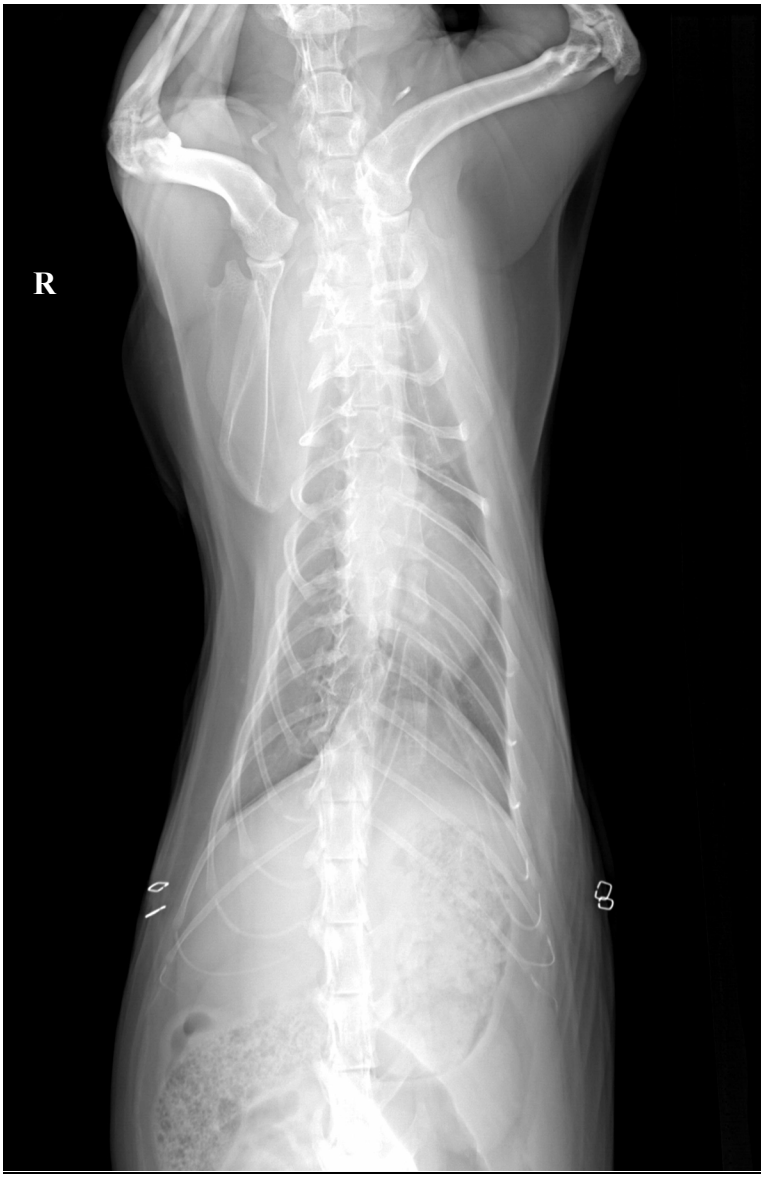


Figure 4b – Thoracic Radiographs Seven Days After Thoracostomy Tube Removal

Ventrodorsal



The surgical staples were removed and a complete blood count was sent to the reference laboratory [(b)] (Table 10). All values were within the reference range. The client was instructed to continue administering the amoxicillin-clavulanic acid [(p)] and to return for a recheck in one month.

The cat presented one month later for a recheck examination. His owner reported that he was doing very well, with good appetite and energy. He continued to experience soft stools as well as intermittent vomiting. On physical examination, his temperature was within normal limits (38.0° C) and his heart and lungs ausculted within normal limits. His respiratory rate was 28 breaths per minute, and his weight was 5.96 kg. No recheck radiographs or laboratory samples were obtained. The client was instructed to complete the final week of amoxicillin-clavulanic acid [(p)]. In addition, a probiotic [(r)] was dispensed (one packet with food q24h for five days) to be started after completion of the amoxicillin-clavulanic acid [(p)]. Phone communication with the client two weeks later confirmed that the diarrhea and vomiting had resolved once the antibiotics had been discontinued, and that the cat was doing very well clinically.

Table 10 – Complete Blood Count One Week After Thoracostomy Tube Removal

| Test | Laboratory Value | Reference Range |
|---|---|---|
| WBC | 13.1 K/μL | 4.2 – 15.6 |
| RBC | 7.30 M/μL | 6.0 – 10.0 |
| HGB | 10.8 g/dL | 9.5 - 15 |
| HCT | 34.9% | 29 - 45 |
| MCV | 48 fL | 41 - 58 |
| MCH | 15.3 pg | 11.0 – 17.5 |
| MCHC | 32.1 g/dL | 29 - 36 |
| Neutrophil Seg | 57% | 35 - 75 |
| Lymphocytes | 35% | 20 - 55 |
| Monocytes | 4% | 1 - 4 |
| Eosinophils | 3% | 2 - 12 |
| Basophils | 0% | 0 - 1 |
| Platelets | 223 K/μL | 170 - 600 |
| Absolute Neutrophil Seg | 7,467 /μL | 2,500 – 12,500 |
| Absolute Lymphocytes | 4,585 μL | 1,500 – 7,000 |
| Absolute Monocytes | 524 /μL | 0 - 850 |
| Absolute Eosinophils | 393 μL | 0 – 1,500 |
| Absolute Basophils | 0 μL | 0 - 100 |

*Slide reviewed microscopically; RBC morphology appears normal*

Discussion

This case describes an eight-year-old male-neutered domestic shorthair cat presenting with characteristic signs of pyothorax. At initial presentation, the cat's hemoglobin saturation status should have been evaluated prior to pursuing diagnostics, and supplemental oxygen provided if the hemoglobin saturation was less than 90% on room air. In addition, there were two additional components of the minimum database that should have been acquired. A complete urinalysis should have been performed prior to administration of IV fluids (assuming the patient was stable enough for cystocentesis) to assess renal function and hydration status, and a blood smear should have been examined to look for signs of bands and neutrophil toxicity and to confirm platelet numbers to accompany the in-house complete blood count. In addition, after collection of the thoracic effusion, an in-house fluid analysis should have been performed to measure the total protein of the fluid to confirm that the fluid was an exudate, although the gross appearance was highly suggestive.

The cat appeared clinically dehydrated at initial presentation, with decreased skin turgor and tacky mucous membranes, yet the initial fluid rate was only two mL/kg/hour. This was not appropriately high given the patient's likely hypovolemia and third space losses into the thorax.

The placement of the thoracostomy tubes was not ideal. The left tube was not positioned as cranially as desired, and the right tube appeared to cross the mediastinum to the left side (the heaviest exudate appeared to be on the right side of the thorax). However, cats with pyothorax are often unstable during anesthesia, and this patient was experiencing low systolic blood pressure despite crystalloid and colloid boluses. More aggressive anesthetic techniques, such as the administration of pressor therapy or changing from isoflurane to a balanced anesthesia protocol, could have been utilized to stabilize blood pressure in order to prolong anesthesia. The decision to leave the thoracostomy tubes in position and to recover the patient seems appropriate in retrospect, however, given the fact that both tubes appeared to be productive during flushing, thoracostomy tubes often are not ideally placed, and the highest mortality risk is associated with the first 48 hours of hospitalization.

There were two areas in which supportive care could have been improved: patient monitoring while on dopamine and caloric intake. After the thoracostomy tubes were placed on day one, the cat was hospitalized overnight under

observation by registered veterinary technicians, but blood pressure and heart rate were not monitored while the cat was on a continuous rate infusion of dopamine. Blood pressure and heart rate should have been monitored at one- to two-hour intervals overnight. The patient could have been hypo- or hypertensive, or experienced a tachycardia or an arrhythmia, and this would not have been detected or corrected. In addition, during days two through seven, the cat was not eating well and was losing weight. While the patient did eventually begin to eat, more attention should have been paid to daily caloric requirements versus estimated daily caloric intake, and careful observations about food preferences noted when the cat did eat. They delayed use of an appetite stimulant (added on day six) was a clinical error.

There were several areas to improve case management in the area of radiographic imaging. Dorsoventral and lateral thoracic radiographs were taken shortly after presentation. It may be preferable for cats in respiratory distress to obtain a single dorsoventral view to confirm the presence of pleural effusion. Thoracic radiographs were obtained on day one and on day three, but were not obtained prior to removal of the thoracostomy tubes on day seven, nor prior to discharge to the owner later in the day. The decision to remove the thoracostomy tubes was based on the absence of significant exudate from the thoracostomy tubes during flushing, observed clinical improvement (normal respiratory rate and effort, normal body temperature, eating and drinking) and improving leukocytosis. However, it is possible that the decrease in aspirated flocculent material was due to pocketing of the exudate rather than resolution of the pyothorax. If the pyothorax had not resolved, the cat could have relapsed after removal of the thoracostomy tubes. Thoracic radiographs were obtained at a recheck one week after discharge, but the ventrodorsal view was moderately rotated – this could have obscured a focal area of bronchopneumonia, lung lobe consolidation or pocketed exudates. This radiograph should have been retaken, since it was used to support the assessment that the pyothorax was clinically resolved. Finally, thoracic radiographs should have been taken at the final recheck examination in week five to confirm resolution of the pyothorax, since antibiotics were discontinued at the end of week six. The pet could have been subject to a relapse by premature withdrawal of antibiotics.

Summary

An eight-year-old male neutered domestic short hair presented for lethargy and inappetence. On examination, the cat was noted to have a restrictive breathing pattern and muffled lung sounds. Diagnostic imaging showed that the cat had pleural effusion. Thoracocentesis was performed, and cytologic findings were consistent with a pyothorax. Bilateral thoracostomy tubes were placed under general anesthesia, and the cat was hospitalized for seven days for supportive care. Aerobic and anaerobic culture and sensitivity showed that the cat was infected with *Actinomyces* spp and *Fuosobacterium* spp. Thoracostomy tubes were removed on day seven, and the cat was discharged on a six-week course of amoxicillin-clavulanic acid. (p) The cat was disease-free based on radiographs taken one week after discharge, and did not relapse at the completion of antibiotics. The prognosis for cats with pyothorax medically managed with thoracostomy tubes is good if they survive the first 48 hours after initial presentation.

Endnotes

a. MyLab™ 50, Esaote, Genova, Italy

b. IDEXX Reference Laboratories, Westbrook, Maine, USA

c. VetTest® Chemistry Analyzer, IDEXX Laboratories, Westbrook, Maine, USA

d. LaserCyte® Hematology Analyzer, IDEXX Laboratories, Westbrook, Maine, USA

e. SNAP® FIV/FeLV Combo Test, IDEXX Laboratories, Westbrook, Maine, USA

f. VetScan i-STAT 3 Analyzer, Abaxis, Union City, California, USA

g. Normasol® R, Abbott Laboratories, North Chicago, Illinois, USA

h. Timentin®, Ticarcillin Powder for Injection, GlaxoSmithKline, Middlesex, United Kingdom

i. Doppler Ultrasonic Flow Detector 811-B, Parks Medical Electronics, Aloha, OR, USA

j. PropoFlo™ Injectable, Abbott Animal Health, Abbott Park, Illinois, USA

k. Cardell Veterinary Monitor 9405, Midmark, Tampa, FL, USA

l. Argyle™ Trocar Catheter, 8 French (2.7 mm) x 9" (23 cm), Covidien, Mansfield, Massachusetts, USA

m. 6% Hetastarch, Teva Parenteral Medicines Inc., Irvine, California, USA

n. Intensive Care Unit, Snyder Mfg. Co., Centennial, Colorado, USA

o. Metacam® Oral Suspension, 1.5 mg/mL, Boehringer Ingelheim, Ingelheim am Rhein, Germany

p. Clavamox® (amoxicillin trihydrate, clavulanate potassium), Pfizer Animal Health, St. Louis, Missouri, USA

q. TELAZOL, Teletamine HCl and Zolazepam HCl, Fort Dodge Animal Health, New York, New York, USA

r. Fortiflora®, Purina Veterinary Diets, Ralston Purina Company, St. Louis, Missouri, USA

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12. Doyle RS, Bellenger CR, Campoy L, et al. Pyothorax in a cat managed by intrathoracic debridement and postoperative ventilatory support. Irish Veterinary Journal 2005;58:211-215.
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