Compendium

Nosocomial Infections

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Abstract: *Nosocomial infections (NIs) are infections acquired during hospitalization. They are characterized by a high incidence of antimicrobial resistance. The most common NIs are pneumonia and urinary tract, surgical site, and bloodstream infections. Hand hygiene has demonstrated efficacy in reducing NIs.*

Nosocomial infections (NIs) are infections acquired by patients during hospitalization. An estimated 5% to 10% of human patients admitted to hospitals develop an NI.[1] Among identified pathogens in human intensive care units (ICUs), 70% are resistant to at least one antimicrobial.[2] In 2008, 64% of biosecurity experts at veterinary teaching hospitals believed that the risk of NI among their patients had increased in the preceding 10 years.[3] Between 2003 and 2008, 82% of veterinary teaching hospitals reported outbreaks of NIs and 45% reported more than one NI outbreak.[3] In human medicine, urinary tract infections (UTIs), pneumonia, surgical site infections (SSIs), and bloodstream infections (BSIs) account for approximately 80% of all NIs.[4] This article reviews the most common NIs, the human and veterinary literature for each type of infection, and the diagnostic and treatment protocols as well as prevention strategies.

Urinary Tract Infections

UTIs are the most common NIs in human hospitals. They account for up to 40% of all human NIs[5] and are typically associated with the placement of a urinary catheter during hospitalization, resulting in catheter-associated UTIs (CAUTIs). Many patients with nosocomial bacteriuria are asymptomatic, and these patients are of concern because they are a major reservoir of antimicrobial-resistant organisms.[6]

The frequency of CAUTI in veterinary studies varies from 10% to 38% of hospitalized dogs.[7–11] However, differences between studies with regard to the signalment of enrolled animals, duration of catheterization, and use of antimicrobials make comparison of results difficult.[7–11] Studies in small animal patients in 1985[12] and 1988[8] demonstrated that even when a closed collection system is used, bacteriuria develops in 32% to 52% of cases. A more recent study[13] found that the incidence of nosocomial bacteriuria in dogs with open urine collection systems was not significantly different from those in dogs with closed systems for short periods of catheterization, provided that a strict hygiene protocol was practiced for placement and management of the urinary catheter.

Pathogenesis

Normally, the length of the urethra and unidirectional flow of urine prevent upward migration of microorganisms into the bladder. In addition, the urinary mucosa secretes inhibitors of bacterial adhesion, preventing attachment of pathogens. Several characteristics of urine, including osmolality, pH, and the presence of organic acids, inhibit the growth of microorganisms. The use of urinary catheters interferes with these defense mechanisms, allowing pathogens to colonize the urinary tract[14,15] by ascending into the bladder on either the extraluminal or intraluminal surface of the catheter.[16] Microorganisms may enter

Key Points

- A diagnosis of CAUTI can only be made on the basis of sterile cystocentesis, as a study demonstrated poor agreement between culture results of urine samples and urinary catheter tips.
- Use of histamine blockers and proton-pump inhibitors increases gram-negative colonization of the oropharyngeal tract, increasing the risk of hospital-acquired pneumonia due to these organisms in people.
- Infection rates in humans nearly double with every hour the patient spends in surgery.
- IV catheters should be removed as early as medically indicated, but routine catheter changes should be avoided unless there is evidence of an infection.
- Studies have shown a temporal relationship between improved hand hygiene and decreased infection rates.
- Less than 50% of small animal veterinarians and less than 20% of large animal and equine veterinarians wash their hands between patient contacts.

Box 1. Criteria for Diagnosis of Catheter-Associated Urinary Tract Infections in Humans[4]

1. Positive urine culture growing >10^5 colony-forming units/mL, with no more than two microorganism species

and

Signs of urinary tract infection:

- Fever
- Urgency
- Frequency
- Dysuria

or

2. Two of the following signs:

- Fever
- Urgency
- Frequency
- Dysuria

and

One of the following laboratory findings:

- Positive gram stain of a urine sample
- Pyuria (>3 white blood cells/high-power field)
- Two urine cultures with >10^2 colony-forming units/mL of a single pathogen in a patient being treated with antimicrobials

Box 2. Indications for Placement of Urinary Catheters in Humans[25,a]

- Accurate monitoring of urine output in a critically ill patient
- Acute anatomic or functional urinary retention or obstruction
- Perioperative use for selected surgical procedures (long anticipated duration of surgery, urologic procedures, need for intraoperative monitoring of urine output)
- Urinary incontinence in patients with open wounds that may be contaminated with urine
- Patient comfort for end-of-life care

[a]Urinary incontinence alone is not an indication for catheterization.

the bladder extraluminally either at the time of catheter insertion or by ascending the mucous film surrounding the external aspect of the urinary catheter and are typically endogenous to the patient, arising from the rectum or perineum. Alternatively, microorganisms may migrate intraluminally, which typically occurs when the internal lumen of the catheter is colonized either through failure of a closed drainage system or contamination of the drainage bag. Bacteriuria in this setting often involves multidrug-resistant organisms. In one human study,[16] extraluminal migration was most likely in two-thirds of cases of NI, with intraluminal migration most likely among the remainder.

Biofilms are composed of clusters of microorganisms and extracellular matrix (primarily polysaccharide materials) and form readily on the extraluminal and intraluminal surfaces of urinary catheters.[15] Biofilms are typically composed of only one type of microorganism, although polymicrobial biofilms are possible.[17] Antimicrobials tend to penetrate poorly into biofilms, and microorganisms grow more slowly in biofilms, rendering many antimicrobials less effective.[18,19]

Diagnosis

According to the National Healthcare Safety Network, there are two sets of diagnostic criteria for CAUTI (**BOX 1**).[4] However, the diagnostic sensitivity is questionable because symptoms associated with UTI are reported in only 10% of humans with CAUTI.[6] Fever is common in the ICU, but UTI is rarely the cause.[20] Pyuria is also not a reliable indicator of UTI in the setting of catheterization because up to 30% of catheterized patients have pyuria, even in the absence of bacteriuria.[21]

Diagnosis of CAUTI in veterinary patients can only be made on the basis of sterile cystocentesis, as a study demonstrated poor agreement between culture results from urine collected via a sterile infusion plug and those from urine collected from urinary catheter tips.[7] Both antimicrobial-sensitive[7] and antimicrobial-resistant[8,9] organisms have been identified in CAUTI in various veterinary studies, and adjustment of antimicrobials should be dictated by culture results. The use of systemic antimicrobials during catheterization of small animals may decrease the frequency of CAUTIs, but the infections that develop tend to have increased antimicrobial resistance.[22]

Treatment

In humans, bacteriuria commonly resolves spontaneously after urinary catheter removal; however, it can persist and lead to a UTI. Consequently, humans are screened for persistent bacteriuria 48 hours after catheter removal, and treatment is initiated if bacteriuria persists.[23] Because of the presence of biofilm, leaving the catheter in place makes it difficult to eradicate bacteriuria and can lead to the development of antimicrobial resistance.[24] Therefore, the urinary catheter must be removed when treating a CAUTI.

Strategies for Prevention

The most effective strategy for prevention of CAUTIs is avoidance of urinary catheterization unless absolutely necessary.[25] Appropriate indications for urinary catheter placement in humans are summarized in **BOX 2**[25]; these indications may also be applicable to veterinary species. Inappropriate use of urinary catheters in human hospitals is reported in up to 50% of hospitalized patients.[26,27] Such an evaluation has not been performed in veterinary patients, although it is reasonable to assume that overuse of urinary catheters occurs in the veterinary setting as well.

CAUTIs can also be minimized by limiting the duration and frequency of catheterization and adhering strictly to aseptic technique and hygiene. Breaks in aseptic technique during catheter placement and disruption of the closed system are the most significant factors in the development of CAUTIs.[20] The collection system should not be raised above the level of the patient, and the collecting lines should not be flushed because urine in the line and bag must be considered contaminated. The collecting bag should remain below the level of the bladder to prevent reflux of urine

Box 3. Conditions That Increase Risk of Aspiration Pneumonia in Small Animals[33–45]

- Laryngeal or esophageal disorders
- Decreased mentation or recumbency from neurologic disease
- Recent sedation or anesthesia
- Long-distance physical exertion
- Use of feeding tubes

into the bladder and should be emptied routinely. Unobstructed urine flow should be maintained at all times.[28]

Pneumonia

Hospital-acquired pneumonia (HAP) is defined as pneumonia that develops more than 48 hours after hospital admission in the absence of any signs of infection at the time of admission.[29] HAP may increase a human patient's hospital stay by more than a week and mortality by three-fold.[29] HAP is 20 times more likely to occur in ventilated patients than in nonventilated patients and can occur in up to one-third of patients requiring mechanical ventilation.[29]

Pathogenesis

The pathogenesis of HAP is multifactorial. Severe illness and hemodynamic compromise have been associated with increased rates of HAP.[30] Supine position greatly increases aspiration risk and has been demonstrated to increase the rate of HAP among hospitalized human patients.[31] Use of gastric ulcer prophylaxis such as histamine blockers and proton-pump inhibitors is associated with increased gram-negative colonization of the oropharyngeal tract, increasing the risk of HAP in people.[29] Endotracheal and nasogastric tubes also increase the risk of HAP by acting as physical conduits for the migration of pathogens to the lower respiratory tract.[29]

Only one study[32] examining nosocomial pneumonia—as a complication of positive-pressure ventilation (PPV) in cats—has been reported in the veterinary literature. In this study, pneumonia was identified in 14 cats, eight of which fulfilled the criteria for ventilator-associated pneumonia (VAP). The most common organisms identified included *Escherichia coli* (10) and *Acinetobacter* spp (6), and multiple organisms were identified in approximately half of the cases. The authors did not differentiate between organisms identified in patients with pneumonia and in patients with VAP, and susceptibility testing was not reported in this study. The incidence of VAP was significantly higher in survivors than in nonsurvivors, which the authors attributed to the length of time spent on positive-pressure ventilation.[32]

Although aspiration pneumonia does not meet the strict definitions of HAP in people, the incidence of aspiration pneumonia in postoperative hospitalized dogs has been reported in a variety of studies.[33–37] However, most of these studies did not examine the effect of aspiration pneumonia on morbidity and mortality. Conditions described in the veterinary literature that increase the risk of aspiration pneumonia are shown in **BOX 3**; some are similar to risk factors for HAP in people.[33–45] A recent study on aspiration pneumonia in dogs[46] reported that half of the patients were receiving H_2 blockers for gastric ulcer prophylaxis, although it is unclear what role the use of H_2 blockers played in the development of aspiration pneumonia. The most common organisms identified were *Mycoplasma*, *Pasteurella,* and *Staphylococcus* spp, as well as *E. coli*; antibiotic sensitivities were not reported. The survival rate for dogs with aspiration pneumonia has been reported to be good in two retrospective studies from veterinary academic facilities.[46,47]

Box 4. Centers for Disease Control and Prevention/National Healthcare Safety Network Criteria for Probable Hospital-Acquired Pneumonia[4,29]

Two or more serial chest radiographs (in patients without underlying pulmonary or cardiac disease, one definitive chest radiograph is acceptable) with at least one of the following:

- New or progressive and persistent infiltration
- Consolidation
- Cavitation

and

At least one of the following clinical criteria:

- Fever with no other recognized cause for fever
- Leukopenia or leukocytosis
- Altered mental status with no identifiable cause

and

At least two of the following criteria:

- New onset of purulent sputum, change in character of sputum, increased respiratory secretions, or increased suctioning requirements
- New onset or worsening cough, or dyspnea, or tachypnea
- Rales or bronchial breath sounds
- Worsening gas exchange, increased oxygen requirements, or increased ventilator demand

Diagnosis

Based on current human guidelines for the identification of HAP (**BOX 4**), HAP should be suspected in any patient that develops depression, fever, leukocytosis, and cough or dyspnea after periods of vomiting or intubation.[4,29] However, clinical findings such as fever, leukocytosis, and purulent secretions are known to occur in noninfectious pulmonary conditions (e.g., atelectasis, acute respiratory distress syndrome) in people; therefore, they lack specificity for the diagnosis of HAP.[48,49] Similarly, findings on chest radiographs can be nonspecific, as a study found that no

Table 1. Criteria for Defining a Surgical Site Infection in Humans[65]

Factor	Infection Location		
	Superficial Incision	Deep Incision	Organ/Space
Depth of infection	Skin or subcutaneous tissues of incision	Deep soft tissues (fascial and muscle layers)	Any part of anatomy (organ or space) other than incision that was opened or manipulated during operation
Onset of infection in relation to surgery	Within 30 days of surgery	Within 30 days of surgery with no implant **or** within 1 year if implant is in place and infection appears to be related to operation	Within 30 days of surgery with no implant **or** within 1 year if implant is in place and infection appears to be related to operation
Characteristics	• Purulent drainage • Organisms isolated from culture of aseptically obtained fluid or tissue • Signs of infection (pain, tenderness, swelling, heat)	• Purulent drainage, but not from organ/space component of surgical site • Deep incision spontaneously dehisces or is deliberately opened by a surgeon when patient has fever, localized pain, or tenderness • Abscess or other evidence of infection diagnosed by direct examination, during reoperation, or by histopathologic evaluation	• Purulent drainage from a drain that is placed through a stab wound into organ/space • Organisms isolated from culture of aseptically obtained fluid or tissue • Abscess or other evidence of infection involving organ/space that is found on direct examination, during reoperation, or on histopathologic evaluation

radiographic sign correlated well with the presence of pneumonia in mechanically ventilated humans.[50] Air bronchograms were the only radiographic sign that correlated with autopsy-verified pneumonia, but this sign correctly predicted only 64% of cases.[50]

Treatment

When pneumonia is suspected, a sample of bronchial secretions should be obtained and empiric antimicrobial therapy initiated until antimicrobial sensitivity results are available. In veterinary patients, it is imperative to collect samples from the pulmonary parenchyma, as one study found bacterial organisms identified on deep oral swabs are inconsistent with organisms identified in tracheal wash samples in dogs.[51] Empiric treatment with third- or fourth-generation cephalosporins, monobactams (aztreonam), piperacillin-tazobactam, or imipenem-cilastatin is recommended in human patients with nosocomial pneumonia.[52] This strategy has been shown to improve outcome in human studies but eventually promotes colonization by multidrug-resistant pathogens.[53]

It is important for clinicians to recognize that the predominant pathogens associated with hospital-acquired infections may vary between hospitals as well as among specialized units within the same hospital.[54,55] Consequently, routine surveillance is recommended to determine the most common nosocomial pathogens in individual hospitals.[54,55] Failure to treat VAP with an appropriate initial antimicrobial regimen has resulted in significantly higher rates of septic shock and hospital mortality in people.[56–58] Additionally, treatment delays of >24 hours after identifying diagnostic criteria for VAP have been associated with statistically higher rates of bacteremia and in-hospital mortality.[59] Rapid diagnosis and institution of therapy are critical to a successful outcome for patients with HAP.

Surgical Site Infections

SSIs are the third most common type of NI in human medicine,[60] prolonging hospitalization and contributing significantly to the morbidity and mortality of affected human patients.[61] The duration of the surgical procedure has been cited as the most important contributor to the development of SSIs in people and animals, with infection rates in humans nearly doubling with every hour the patient spends in surgery.[62,63] A veterinary study found that nosocomial SSIs increased the duration of postoperative and total hospitalization.[64] The Centers for Disease Control and Prevention has developed standardized criteria for diagnosing SSIs in people (**TABLE 1**).[65]

Pathogenesis

Microbial contamination of the surgical site is a necessary precursor of SSI. It has been shown that if a surgical site is contaminated with $>10^5$ microorganisms per gram of tissue, the risk of SSI is markedly increased.[66] For most SSIs, the source of pathogens is endogenous flora of the patient's skin, mucous membranes, or hollow viscera.[67] Gram-negative bacteria produce endotoxin, which stimulates cytokine production and can trigger systemic inflammatory response syndrome, resulting in multiple organ dysfunction.[68,69] Gram-positive bacteria produce glycocalyx and an associated component called *slime*, which physically shields the bacteria from phagocytes or inhibits the binding or penetration of antimicrobial agents.[70–72]

Risk Factors

A comparison of risk factors for SSI in people and veterinary patients is listed in **TABLE 2**.[62–65,73–79] In veterinary species, intact male status was identified as a risk factor for SSIs, which was speculated to be

Table 2. Risk Factors for Surgical Site Infections in Human Versus Veterinary Patients[62–65,73–79]

Factor	Human Patients	Veterinary Patients
Patient characteristics	• Extremes of age • Poor nutritional status • Diabetes (controversial) • Smoking • Obesity • Coexistent infections at a remote body site • Colonization with microorganisms • Blood transfusion (due to transfusion-related immunosuppression)	• Intact male • Endocrinopathy —Diabetes mellitus —Hyperadrenocorticism —Hypothyroidism • Increasing body weight
Surgical procedure factors	• Inadequate duration of surgical scrub • Inadequate skin antisepsis • Premature preoperative shaving • Premature preoperative skin preparation • Prolonged duration of procedure • Poor antimicrobial prophylaxis • Inadequate operating room ventilation • Inadequate sterilization of instruments • Foreign material at surgical site • Poor surgical technique • Poor hemostasis • Failure to obliterate dead space • Tissue trauma	• Increased time between surgical site preparation and surgery • Prolonged duration of surgery • Prolonged duration of anesthesia independent of surgical time • After-hours versus daytime surgery • Inappropriate antimicrobial therapy • Increased number of people in operating room • Drain placement

related to depressed cytokine production in intact males compared with castrated males.[78] In another veterinary study, concurrent endocrinopathies were also associated with an increased risk of SSIs due to depressed natural killer cell and lymphocyte number and function in animals with endocrine disorders.[74] Prophylactic antimicrobials are indicated in many surgical procedures, but they should be limited to the immediate perioperative period in most cases. A study[64] found that dogs receiving perioperative antimicrobials when subjected to clean-contaminated surgical procedures were six to seven times less likely to develop an SSI than patients without antimicrobial prophylaxis. However, another study concluded that the postoperative infection rate was increased in small animals receiving prolonged postoperative antimicrobials compared with those receiving only perioperative antimicrobials.[73] First-generation cephalosporin antimicrobials are commonly selected for perioperative use because they have excellent activity against *Staphylococcus* spp and *E. coli*. They also have minimal toxicity and are beneficial for use in the perioperative period.[64]

Treatment

Coagulase-positive staphylococci are the most common isolates in reports of SSIs in small animal patients.[77] Samples for culture should be collected at the time of definitive therapy for the SSI, particularly if the patient is already receiving antimicrobials, as the possibility of the development of a multidrug-resistant organism increases with previous antimicrobial use. The goal of surgery in the treatment of an SSI should be to reduce the load of microorganisms, remove necrotic tissue, and maintain adequate tissue perfusion.[79]

Prevention

In human medicine, shaving before surgery is advised against because any method of hair removal can damage the epithelium, allowing bacterial colonization, and shaving has been shown to increase SSI rates.[80] Leaving hair and fur in the surgical field is not a viable option for most veterinary patients, but clipping the surgical site should be performed as late as possible, as shaving the night before surgery has been associated with higher SSI rates in humans.[81,82] Similarly, a study in dogs and cats found higher SSI rates when clipping was performed before induction of anesthesia compared with after induction.[73] In addition, the clipper blades should be cleaned and, ideally, sterilized between uses because more frequent use without sterilization increases bacterial colonization of clipper blades.[83]

Because duration of surgery is an important risk factor for the development of SSIs in veterinary patients, short surgical times are essential to reduced SSI complication rates.[64,73,74] However, poor surgical technique is associated with increased SSI rates in people.[65] Consequently, every effort must be made to keep surgical times as brief as possible without compromising quality of technique.

Bloodstream Infections

The incidence of BSIs in human hospitals has steadily increased in the past 2 decades, and most BSIs are related to intravascular devices, particularly central venous catheters (CVCs).[84] BSIs are associated with a high fatality rate, exceeding 25% in some reports.[85] Duration of catheterization is the most important risk factor for the development of catheter-related BSIs (CR-BSIs),[86] with most infections developing after 4 to 5 days.[87]

Bacterial contamination of catheters in critically ill animals has been speculated to increase morbidity and mortality rates, as bacterial colonization is considered a precursor to catheter-related infection.[88] In a study of 88 critically ill dogs, the incidence of bacterial colonization of IV catheters ranged from 15% to 48%,

Table 3. Mechanism and Spectrum of Activity of Antiseptic Agents Commonly Used for Preoperative Skin Preparation and Surgical Scrubs[97–100]

Agent	Mechanism of Action	Gram-Positive Bacteria	Gram-Negative Bacteria	Fungi	Viruses	Rapidity of Action	Residual Activity	Toxicity
Alcohol	Denature proteins	Excellent	Excellent	Good	Good	Most rapid	None	None
Chlorhexidine	Disrupt cell membrane	Excellent	Good	Fair	Good	Intermediate	Excellent	Ototoxicity, keratitis
Iodine/ iodophors	Oxidation/ substitution by free iodine	Excellent	Good	Good	Good	Intermediate	Minimal	Absorption from skin with possible toxicity, skin irritation

and the most common organisms isolated included *E. coli* and *Aerobacter*, *Proteus*, and *Klebsiella* spp.[88] Another study of animals in a small animal ICU reported that 26% of jugular catheters were positive for bacterial growth; enteric organisms were most commonly isolated.[8] Two other studies reported colonization rates of 7% in dogs and cats receiving total parenteral nutrition[89] and 22% of dogs with parvovirus.[90] More recently, a study on IV catheters from dogs and cats hospitalized for at least 24 hours in the ICU found a positive culture rate of 24.5%, with *Enterobacter* spp being the most common organisms identified (46%).[91] Several risk factors were examined, including catheter type, location, duration, and blood sampling from the catheter, but none was associated with increased risk of CR-BSI. In human medicine, the organisms most often implicated in CR-BSI are skin commensals, whereas in veterinary patients they are typically enteric and environmental organisms.[8,88–90] Previously, outbreaks of CR-BSIs in veterinary hospitals have been linked to inadequate skin preparation,[88] contaminated gauze squares,[92] and other, unidentified vehicles.[93]

Diagnosis and Treatment

If a catheter infection is suspected, the catheter should be removed using sterile technique, and the tip of the catheter should be submitted for bacterial culture and sensitivity testing in conjunction with blood samples from central and peripheral sites.[94] Initial antimicrobial therapy should be broad spectrum, particularly if a life-threatening bacteremia is suspected. However, veterinary studies have reported a high incidence of resistant organisms colonizing intravenous catheters, characterized by high levels of resistance to penicillin, cloxacillin, erythromycin, and cephalexin.[90] It is rare for CR-BSIs to be associated with inflammatory signs at the insertion site,[95] but when present, these signs are reliable predictors.[96] Clinicians should consider the diagnosis of CR-BSI in patients with fever, hypotension, leukocytosis, or other signs of sepsis. A definitive diagnosis is made when the same organism is cultured from a percutaneous blood sample and the catheter tip.[94] The antimicrobial selection should be narrowed when culture results are available.[94]

Prevention

A number of studies have attempted to determine the optimal agent for skin cleansing before CVC insertion and at times of CVC manipulation. Chlorhexidine is thought to have a theoretical advantage over povidone-iodine because it has a prolonged time of antimicrobial effect and because it is not inactivated by exposure to protein-rich fluids such as blood and serum.[97,98] A 2002 meta-analysis examined eight randomized trials comparing various types of chlorhexidine and iodine solutions and found that use of chlorhexidine solutions had less than half the risk of catheter colonization and CR-BSI.[99] There is also evidence that alcohol and chlorhexidine may have synergistic activity against bacteria in vitro.[100] **TABLE 3** lists characteristics of commonly used agents for skin cleansing in veterinary patients.[97–100]

A number of studies comparing transparent and gauze dressings in humans have been performed, some showing no difference and others suggesting increased risk of infection with transparent dressings.[101–103] These conflicting results have allowed for continued use of gauze and transparent gauze dressings depending on institutional preferences. The ideal interval between dressing changes depends primarily on the type of dressing used. The use of gauze dressings changed every 2 days appears equivalent to the use of transparent dressings changed every 5 days with regard to rate of colonization and is the most recommended standard of care.[102]

A number of trials have examined whether the use of prophylactic antibiotics at the time of catheter insertion has any effect on infection rates.[104–106] None demonstrated any reduction in episodes of CR-BSI, and a 2005 Cochrane review concluded that there was no role for prophylactic antibiotics at the time of CVC insertion.[107]

Despite the increased risk of infection with prolonged catheterization, studies in human patients have indicated that prophylactic catheter changes every 3 days versus every 7 days did not decrease the incidence of catheter-related bacterial colonization.[108] These studies have led to the current recommendation in human medicine that catheters be removed as early as medically indicated but that routine catheter changes be avoided unless there is

Box 5. Indications for Hand Hygiene[121]

- Before and after touching the patient
- Before handling an invasive device for patient care, regardless of whether gloves are used
- After contact with body fluids or excretions, mucous membranes, nonintact skin, or wound dressings
- If moving from a contaminated body site to another body site during care of the same patient
- After contact with inanimate surfaces and objects (including medical equipment) in the immediate vicinity of the patient
- After removing sterile or nonsterile gloves

evidence of an infection.[108] A prospective veterinary study showed that intravenous catheters can remain in place for more than 3 days (up to 10 days based on study limitations) in a peripheral vein provided that strict aseptic technique is observed during placement and catheter care is vigilant.[92]

Hand Hygiene for Prevention of Nosocomial Infections

Results of human studies indicate that at least one-third of all NIs are preventable.[109] Nosocomial pathogens have been shown to persist in the hospital environment on items such as stethoscopes,[110] computer keyboards, and faucet handles.[111] However, evidence that disinfection of environmental surfaces influences NI rate is lacking. A review of scientific articles and abstracts investigating the effect of environmental disinfection on NI rates failed to demonstrate a relationship between routine disinfection of surfaces (mainly floors) with lower infection rates.[109] We do not recommend that disinfection of environmental surfaces in the hospital be abandoned, but rather that efforts to limit NI should be directed by more proven measures, specifically hand hygiene.

The hands of health care workers (HCWs) are the primary vehicle of transmission of NIs to patients.[112] Therefore, hand hygiene is a key component in the prevention of NI.[112] HCWs can contaminate their hands even by performing so-called "clean procedures," such as lifting a patient; taking a patient's pulse, blood pressure, or temperature; or touching intact areas of a hospitalized patient's skin.[113–115] HCWs may also contaminate their hands after touching inanimate objects.[115–117] Several outbreaks of NIs have been associated with HCWs' hands.[118–120] Indications for hand hygiene are listed in **BOX 5**.[121]

The purpose of routine hand hygiene in patient care is to remove dirt and organic material. *Hand washing* refers to the application of a plain (nonantimicrobial) or antiseptic (antimicrobial) soap. This method of cleaning mechanically removes dirt (soiled and organic substances) and loosely adherent flora from the hands. Plain soaps have minimal or no antimicrobial activity.[122] In contrast to hand washing, alcohol-based hand rubs rapidly reduce skin flora by killing as alcohols denature proteins.[123] Alcohols have excellent in vitro activity against gram-positive and gram-negative bacteria, including methicillin-resistant *Staphylococcus aureus,* vancomycin-resistant enterococci, and a variety of fungi, but they have poor activity against protozoan oocysts, nonenveloped viruses, and bacterial spores.[123–125] A review of effectiveness of alcohol-based solutions for hand hygiene showed that alcohol-based hand rubs remove more organisms more effectively, require less time, and irritate the skin less than hand washing with soap or other antiseptics and water.[126] Consequently, in 2002, the Healthcare Infection Control Practices Advisory Committee Guidelines defined alcohol-based hand rubs as the standard of care for hand hygiene in health care settings.[127]

Studies have shown that improving hand hygiene decreases NI rates.[127] However, compliance with hand washing protocols in human hospitals remains poor.[128] This is consistent with a veterinary study that showed that <50% of small animal veterinarians and <20% of large animal and equine veterinarians wash their hands between patient contacts.[129] In addition, sustained improvements in hand washing are difficult and require ongoing monitoring of compliance.[128] Guidance for the implementation of effective hand hygiene campaigns is available at the CDC Web site (www.cdc.gov/handhygiene).

Conclusion

NIs cause a significant increase in morbidity and mortality in human medicine, and awareness of NI is increasing in veterinary medicine. Adherence to recommendations for the prevention, identification, and management of specific NIs can help improve outcomes for veterinary patients. The most important factor in preventing NIs is hand hygiene, which has been shown to dramatically reduce transmission of bacteria between hospitalized patients.

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1. Which is not one of the four most common NIs?

a. UTI
b. SSI
c. meningitis
d. pneumonia
e. BSI

2. What percentage of nosocomial pathogens is resistant to at least one antibiotic?

a. 20%
b. 40%
c. 50%
d. 70%
e. 80%

3. Which is not a risk factor for aspiration pneumonia in small animals?

a. laryngeal or esophageal disorders
b. proton-pump inhibitor administration
c. recent sedation or anesthesia
d. long-distance physical exertion
e. use of feeding tubes

4. Which of the following is not one of the most common microorganism species identified in a recent study on aspiration pneumonia in dogs?

a. *Mycoplasma*
b. *Pasteurella*
c. *Staphylococcus*
d. *Escherichia*
e. *Streptococcus*

5. Which is not a risk factor for SSIs in human or veterinary medicine?

a. drain placement
b. prolonged duration of surgery
c. increased time between surgical site preparation and surgery
d. use of ketamine
e. increased number of people in the operating room

6. Which of the following has been associated with the development of CR-BSIs in veterinary patients?

a. IV catheter location
b. IV catheter type
c. duration of IV catheterization
d. blood sampling from an IV catheter
e. none of the above

7. Which is reported to be the best skin cleansing solution when preparing for IV catheter placement?

a. chlorhexidine
b. povidone
c. alcohol
d. soap and water
e. none of the above

8. Which practice has been shown to be most effective at reducing NIs?

a. hand hygiene
b. disinfection of surfaces
c. use of bleach
d. wearing protective clothing
e. isolation of possibly contagious patients

9. What is the reported compliance rate of hand washing among small animal veterinarians?

a. <20%
b. <40%
c. <50%
d. <70%
e. <90%

10. Which is the recommended method of hand hygiene in human medicine?

a. use of alcohol-based hand solutions
b. use of soap and water
c. use of water alone
d. hand washing is not recommended
e. a and b are equally recommended