

Source: Journal of Dental Hygiene, Vol. 79, No. 3, Summer 2005

Copyright by the American Dental Hygienists' Association

Cross-contamination of the PSP Sensor in a Preclinical Setting

Wendy Negron, Sally M Mauriello, Charlotte A Peterson and Roland Arnold

Wendy Negron, BSDH, MS, is manager of clinical and professional relations at Omnii Oral Pharmaceuticals. Sally M. Mauriello, EdD, is associate professor; Charlotte A. Peterson, MS, is clinical associate professor; and Roland Arnold, PhD, is professor; all are at the University of North Carolina School of Dentistry in Chapel Hill.

Purpose. *This study tested an infection control protocol for photostimulable phosphor (PSP) sensors while simulating multiple uses.*

Methods. *Dental hygiene students (n=36) were randomly assigned to a control group (dry wipe) or test group (disinfectant wipe). A sterile, barriered sensor was placed in a peer patient's mouth to simulate an exposure. After simulation, the barriered sensors were disinfected, placed into a new barrier, and inserted into the same peer patient's mouth. Following the fourth placement and treatment, sensors were vortexed in specimen cups containing trypticase soy broth (TSB) and incubated at 37C to detect contamination by oral bacteria.*

Results. *Bacterial growth in TSB was monitored as a change in optical density and by quantitation of oral streptococci on Mitis-salivarius agar. The barrier-protected sensors were minimally contaminated (<10⁵ colony forming units/ml at baseline) regardless of treatment. After culture amplification, the control TSB had more samples that remained negative through 48 hours (5/18 vs. 0/18 for test group, p<0.05).*

Conclusion. *Barrier envelopes used with the PSP sensors appeared to be an effective way of reducing microbial contamination.*

Keywords: Infection control, cross-contamination, barrier-protected sensor, oral radiography, microbial contamination

Introduction

Previous studies on film-based imaging have shown the need to exercise universal precautions in intraoral radiography.¹⁻¹² These findings resulted in a recommendation by the American Dental Association (ADA) and the Centers for Disease Control and Prevention (CDC) for dental health professionals to employ effective disease prevention procedures while providing radiographic dental services.^{1-6, 8-12} Unlike many clinical dental procedures, exposing and processing intraoral radiographs present infection control issues that may not be addressed by standard disinfection and sterilization techniques. Saliva and blood can contaminate a number of surfaces, such as the processor, darkroom surfaces, and radiographic equipment, all of which cannot be easily sterilized or disinfected.^{4, 5, 11, 13} Thus, the prevention and elimination of contamination is necessary for both the radiography operatory and darkroom. In 2001, Bartoloni published infection control recommendations for intraoral film-based imaging, which included wiping the saliva from the film with a paper towel, wearing gloves when opening film packets in the darkroom, using barriers on equipment and film, disinfecting surfaces,

and sterilizing film holders to decrease cross-contamination.¹⁴ Recent protocols have employed one or more of these suggestions, with some techniques being more effective than others in reducing contamination when using conventional dental film.^{2,3,7,9,10,13,15}

The Organization for Safety and Asepsis Procedures (OSAP) also published infection control recommendations to guide the use of surface disinfectants.²¹ OSAP recommended that surfaces be disinfected by a two-step cleaning and disinfecting process referred to as the "spray-wipe-spray" technique. In this technique, any contaminated surface is sprayed with an appropriate surface cleaner, wiped clean, and subsequently sprayed with disinfectant, which is allowed to remain in contact with the contaminated surface for disinfection according to the product's recommendations. For effective surface disinfection, this technique (surface cleaning with subsequent disinfection) must follow the two-step sequence for the indicated contact time in order to eliminate microorganisms from contaminated surfaces.

Through advancements in technology, film-based imaging is slowly becoming an imaging technique of the past, and digital imaging is emerging as the new standard of practice for dental radiographic imaging. This evolution has created new infection control concerns because, unlike film-based imaging, intraoral digital image sensors are used repeatedly from patient to patient. This, in combination with the inability to heat-sterilize the digital sensors, increases the risk of cross-contamination.¹⁶⁻¹⁹ Therefore, a new challenge-preventing cross-contamination when using digital sensors for intraoral radiography-has been created.

To date, three articles and one abstract have been published that address cross-contamination with digital sensors.^{16-18, 20} Common infection control issues addressed in these studies included the inability to sterilize the sensor, difficulty tearing the barrier envelope, and the accidental contamination of the sensors when removing them from the barrier envelope.

The earliest documented study was conducted by Wenzel et al.¹⁸ His concern involved the potential contamination that may occur with the repeated use of the sensor among patients. This investigator hypothesized that the sensor became contaminated when removed from the barrier envelope.¹⁸ One method he tested was to remove the sensor from the barrier envelope and the plastic sheath using sterile tweezers. The procedure was evaluated using both the charged coupled device (CCD) and photostimulable phosphor (PSP) digital sensors. His results indicated that bacterial contamination posed a minor problem in both radiographic procedures.¹⁸

In a second study conducted in 2000, Hockett et al. evaluated the presence of barrier tears after use with a CCD sensor.¹⁶ Results indicated that perforations occurred in 44% to 51% of the plastic sheaths. The author stated that tears may have occurred when placing or removing the CCD sensor from the plastic sheath, causing perforations to occur. When a finger cot was used in conjunction with the plastic sheath to cover the sensors, significantly fewer perforations of the sheath occurred (approximately 6%).¹⁶

In 2000, an additional study conducted by Hubar et al. evaluated six possible ways to insert a CCD receptor covered by a plastic sheath into various film holding devices.¹⁷ Results from this study indicated that four of the six barrier systems exhibited 6% to 83% perforations. The two barrier systems that did not exhibit any perforations were the XCP. that was covered with the plastic sheath only and the plastic sheath in conjunction with the Snap-a ray.¹⁷ Unlike Hockett's study, the sensor used with the finger cot exhibited 5% to 55% perforations.

A published abstract by Peterson et al. also addressed cross-contamination with intraoral digital sensors.²⁰ In this study, the infection control effectiveness of using a barrier envelope and alcohol wipe with a PSP sensor was evaluated. The results indicated that 100% of the sensors were culture positive on *Mitis-salivarius* broth (MSB), with 25% of the disinfected sensors being positive. Results indicate that the manufacturer's recommended protocol for infection control was not adequate for eliminating contamination.

To date, only four studies have been published that address infection control with digital intraoral sensors.^{16-18, 20} Of these four studies, two investigated contamination of PSP sensors. In both studies, the experienced principal investigator solely performed the infection control procedures that were tested, thus minimizing the contamination variability of the sensors.

Wenzel et al. focused on finding a technique that would eliminate the sensor from touching the barrier envelope. On the other hand, Peterson et al. used the manufacturer's recommendation of an alcohol wipe to clean the PSP barrier envelope, but conducted the study in a laboratory setting. Though both studies have researched ways to minimize contamination, none have assessed the effectiveness of infection control techniques in a simulated clinical environment using the PSP sensor multiple times. Therefore, the purpose of this study was to evaluate an infection control protocol for the PSP digital sensor while simulating multiple uses.

Methods and Materials

Sample Population

The sample population consisted of 36 first-year dental hygiene students enrolled in a pre-clinical radiology course at the University of North Carolina at Chapel Hill. Prior to the study, subjects were required to demonstrate a minimum technical competency level when exposing a film-based full mouth series on a DXTTR manikin (DENTSPLY Rinn) using both XCP and Stabe film holding devices. Two film holding devices frequently used in clinical practice were employed in the study: the extension cone paralleling (XCP) (DENTSPLY Rinn, DENTSPLY International) device, and the Greene Stabe (DENTSPLY Rinn) disposable film holding device. The XCP film-holding device, which is made of plastic, aligns the beam to the receptor. The Stabe device, which is made of Styrofoam, does not align the beam to the film. Due to a limited number of radiographic operatories, the study was conducted on two separate days. Prior to the start of the study, subjects participated in an informational session on the clinical use of PSP sensors followed by a demonstration of the placement of the PSP sensor into the manufacturer barrier envelope.

The digital sensors used in the study were the PSP Size 2 DenOptix imaging plates made by DENTSPLY Gendex (DENTSPLY International, Gendex Division). These were comparable to the size and shape of a Size 2 conventional dental film. Each sensor was sterilized prior to its initial use in the study using ethylene oxide and placed aseptically into a barrier envelope. The barrier envelopes used were Size 2 DenOptix Barrier Envelopes (DENTSPLY Gendex, Gendex Dental X-ray Division, Dentsply International) designed for use with the digital sensors. Two infection control protocols were used in the study: the control group wiped the barrier-enveloped sensor with a dry paper towel prior to opening the barrier envelope, and the test group wiped the barrier-enveloped sensor with a dry paper towel and subsequently wiped the barrier envelope with a Prophene (O-phenylphenol) plus disinfectant-sprayed paper towel prior to opening the barrier envelope. The protocol used for this study was reviewed and approved by the University of North Carolina at Chapel Hill School of Dentistry Committee on Research Involving Human Subjects.

Preclinical Procedure

The 36 subjects were randomly assigned to one of two treatment groups. Eighteen were assigned to the control group (wipe barrier envelope with dry paper towel), and 18 were assigned to the test group (wipe barrier envelope with dry paper towel followed by the Prophene plus wipe). Each subject was given a coded paper cup containing a sterile barrier-enveloped sensor. Students were paired as patient and operator to simulate placement of four intra-oral radiographic projections (two anterior and two posterior projections). Written instructions were provided to each subject specifically outlining the infection control steps that were to be used based on the treatment group assignment. Subjects were advised to read the directions prior to the process and follow the specified directions. Due to the insufficient number of XCP film holding devices, Group 1 (control) used the XCP and Group 2 (test) used the Stabe film holding device.

The barrier-enveloped sensors were placed in the film holding device and inserted into the peer patient's mouth to simulate the radiographic exposure with tubehead alignment. No radiation exposure of the sensor occurred. After simulation, the barrier-enveloped sensor was wiped using the assigned infection control protocol and placed in a clean paper cup. Group 2 (disinfectant) needed two cups for this step: one for the dry-wipe step and the other for the disinfectant-wipe step. Subjects then removed their gloves, washed their hands, and donned clean gloves. With clean gloves, subjects tore the barrier envelope at one end and aseptically dropped the PSP sensor from the barrier envelope into another clean paper cup. Finally, student operators removed their gloves, washed their hands, donned clean gloves, and aseptically inserted the same sensors into clean, unused barrier envelopes for placement into the subjects' mouths for the subsequent simulated projection. This

process took place for all four simulated projections performed on the peer partner. On the fourth sensor placement projection, the barrier-enveloped sensor was treated again according to the assigned infection control protocol, but was not removed from the sealed barrier envelope. Each barriered sensor was placed in a coded paper cup with each subject's number and was transported to the laboratory for analysis.

Laboratory Procedures

With clean, gloved hands, the principal investigator removed each PSP sensor from the hand-torn barrier envelope following standard infection control procedures. Each PSP sensor was dropped into a sterile 110 ml specimen cup (Becton Dickinson Labware, Franklin Lakes, NJ) containing 50ml of trypticase soy broth (TSB). The cups were labeled with each subject's number and all lids were secured. Sterile cups were vortexed (mechanically mixed with liquid moving in a circular motion) for 20 seconds and assessed for visual turbidity by the principal investigator. Discernible cloudiness was observed, and the extent of turbidity was estimated on a positive (+), not sure (+/-), and negative (-) scale at 0 time (baseline), 24, and 48 hours. Since this procedure was not limited to oral *streptococci*, ten 1 volumes of culture supernatants were removed with a sterile pipette initially at 0 time and subsequently at 24 hours and spotted on *Mitis-salivarius* (MS) agar (Difco) for colony forming units (CFU). The plates were incubated in a CO₂ incubator at 37C until the CFU of oral *streptococci* were counted.

In addition, at 0, 24, and 48 hours, 100l volumes of TSB from each sample were pipetted into a 96-well microtiter plate in quadruplet. The microtiter plate reader was a spectrophotometer (Molecular Device Corp., CA, VMax kinetic microplate reader) that measures the amount of light (wavelength=660nm) transmitted through each volume as a measure of the absorbance ($A_{660\text{nm}}$). Its function was to detect the growth of all types of bacteria that could grow aerobically at 37C and, unlike MS agar, was not limited to oral *streptococci*.

Follow-up Survey

A week after the study, subjects were placed in a classroom and asked to complete a five-item, open-ended questionnaire that inquired about the use of the PSP sensors in regards to the infection control procedures (Figures 1, 2, 3 4). The principal investigator distributed the survey, with directions on how to complete the survey written at the top of the page. The purpose of the questionnaire was to explore the subjects' experiences and perceptions during the implementation of the infection control protocol.

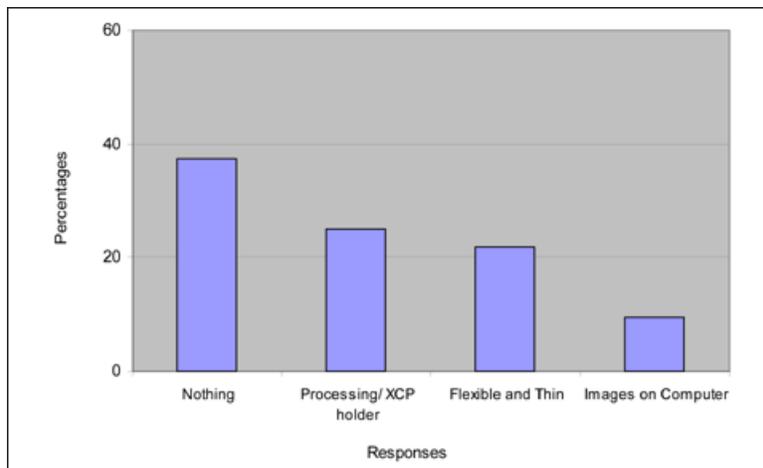


Figure 1: Responses of students when asked: "What did you like most about using the PSP sensor?" (N=32)

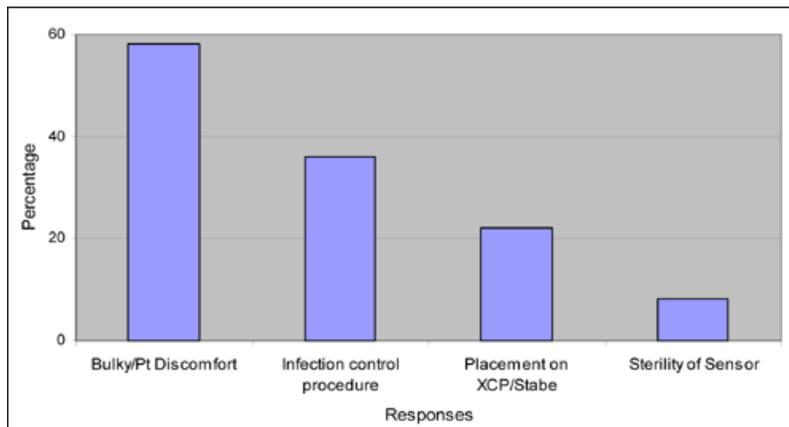


Figure 2: Responses of students when asked: "What do you like least about the PSP sensor?" (N=36)

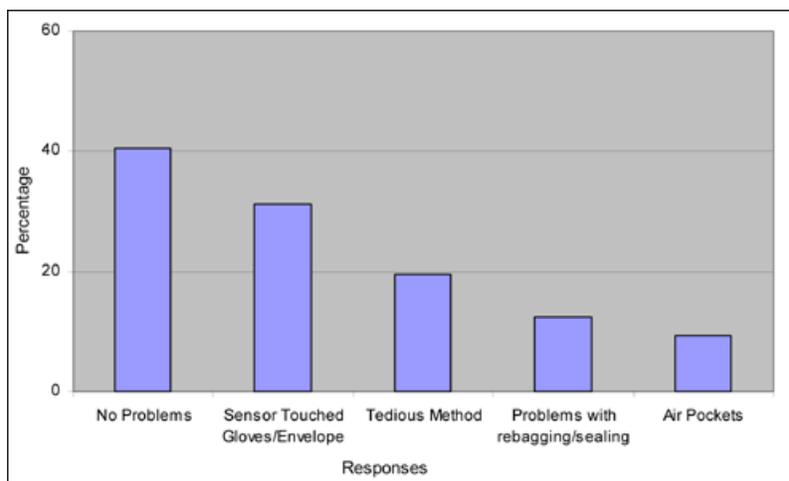


Figure 3: Responses of students when asked: "Describe any problems with infection control procedures." (N=32)

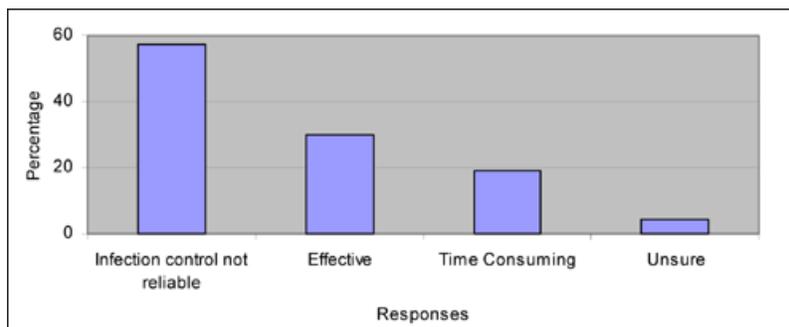


Figure 4: Responses of students when asked: "What was your perception of the infection control protocol used on the PSP sensor?" (N=32)

Statistical Analysis

Visual turbidity is a categorical variable and has meaningful order (-1, 0, 1); therefore, a Mantel-Haenszel chi-square test was performed to determine statistical significance. For optical density and *streptococci* plating, the data did not follow a normal distribution. Therefore, a nonparametric method was applied. A Mantel-Haenszel chi-square test was used to

determine if any statistical difference was observed between groups at each time point. Subject questionnaires were analyzed using frequencies.

Results

A total of 36 first-year dental hygiene students participated in this study, achieving a 100% participation rate. Characteristics of the study population for the dry and disinfectant groups were as follows. Of the 36 subjects in the study, 34 were female and two were male, with 18 females for the dry-wipe group, and 16 females and two males for the wet-wipe group. Twelve of the subjects, seven from the dry-wipe group and five from the disinfectant group, were dental assistants prior to entering the study. All study participants demonstrated the minimum competency performance level of 86% when exposing radiographic film using the XCP film holding device and the Stabe film holding device.

Table I shows the subjective measurement of visual turbidity of PSP sensors from baseline to 48 hours. At baseline, neither group demonstrated a positive visual turbidity test ($p=1.00$). After 24 hours of incubation, Group 1 (dry-wipe) had 27.8% positive sensors, and Group 2 (disinfectant) had 22.2% positive sensors, but no statistically significant difference was found in visual turbidity between groups ($p=0.625$). When examined after 48 hours, bacterial growth was evident by the increased turbidity that had occurred in almost all of the samples. In the dry-wipe group, 72% of the digital sensors resulted in discernible growth in TSB, and in the disinfectant group, 100% of the sensors resulted in visible bacterial growth. The apparent increase in contamination in the disinfectant group approached, but did not reach, significance ($p<0.06$).

Table I. Bacterial growth was assessed as an increase in visual turbidity after incubation of the PSP sensors in TSB for 24h and 48h at 37°C. There was no detectable turbidity immediately after vortexing in any of the samples tested (Baseline).

Time	Group	No Turbidity (-)	Not Sure (+/-)	Positive (+)	Total	P Value
Baseline	1 (Dry)	18	0	0	18	$P=1.00$
	2 (Disinfectant)	18	0	0	18	
Total		36	0	0	36	
24 hours	1 (Dry)	9	4	5	18	$P=0.63$
	2 (Disinfectant)	11	3	4	18	
Total		20	7	9	36	
48 hours	1 (Dry)	4	1	13	18	$P=0.06$
	2 (Disinfectant)	0	0	18	18	
Total		4	1	31	36	

Aliquots of the TSB were sampled at 0, 24, and 48 hours after incubation at 37 C, and the optical densities at 660nm were determined in microtiter format. An $A_{660\text{ nm}} > 0.10$ is visually detectable and was considered positive. Table II presents the contamination status of each sample by time after incubation of the TSB samples. As with visual turbidity, all of the sensors were negative at baseline for bacterial growth. After incubation for 48 hours, five of the 18 dry-wiped sensors remained negative. In contrast, all of the wet-wiped sensors achieved $A_{660\text{ nm}} > 0.10$ by 48 hours. A sensor sample that failed to show a change in optical density at 24 hours but became positive at 48 hours was indicative of less contamination than with a sample that was positive at both 24 and 48 hours. The dry-wipe group consistently had lower sensor contamination than the disinfectant group over 48 hours. The bacterial contamination differed among groups ($p=.02$).

Table II. Bacterial growth was monitored by measuring optical density at 660nm of the TSB containing the PSP sensors at day 0, 1 and 2 after incubation at 37°C. All samples were negative at baseline (0 hour). $A_{660nm} > 0.10$ was considered positive.

Group	Negative at 48h	Negative at 24h, but positive by 48h	Positive by 24h	Total	P Value
Group 1 (Dry)	5	7	6	18	$P=0.02$
Group 2 (Disinfectant)	0	6	12	18	
Total	5	13	18	36	

The change in optical density in TSB would be indicative of any bacterial contamination. A primary concern of the current study was the extent of bacterial contamination that could be attributed to the oral flora. In addition, an increase in optical density is relatively insensitive, requiring a density of $>10^7$ CFU/ml to be considered positive. The fact that optical densities increased after incubation (amplification) for 24 and 48 hours suggested that there was a low level of contamination that was not detectable by optical densities at baseline. Oral *streptococci* dominate the microbial flora of the oral cavity and of saliva and would not be expected as an environmental contaminant. *Mitis-salivarius* agar is selective for oral streptococci and does not permit the growth of environmental contaminants. Table III shows the numbers of TSB samples containing oral *streptococci* between the dry and the disinfectant group at baseline and 24 hours of incubations of the TSB. Only 22.2% and 5.56% of the respective groups (dry-wipe and disinfectant-wipe) had detectable CFU on initial plating. None exceeded 104 CFU/ml. These differences were not significant ($p=0.337$). Following 24 hours of incubation, 38.9% of the samples from the dry-wipe group had no detectable CFU. In contrast, 100% of the sensors from the disinfectant-wipe group had detectable CFU, resulting in statistical difference between groups ($p=0.05$).

Table III. TSB samples containing PSP sensors were plated at time 0 (Baseline) and after 24h incubation for quantitative recovery of oral *streptococci* colony forming units (CFU)/ml on selective MS agar. The limit of detection was 100 CFU/ml.

Incubation Time for TSB	Negative <100 CFU/ml	Positive 10^5 CFU/ml	Positive >math>10^5</math> CFU/ml	Total	P Value
Baseline 0 hr					
Group 1 (Dry)	14	4	0	18	$P=0.34$
Group 2 (Disinfectant)	17	1	0	18	
Total	31	5	0	36	
Day 1 24 hr					
Group 1 (Dry)	7	5	6	18	$P=0.05$
Group 2 (Disinfectant)	0	9	9	18	
Total	7	14	15	36	

Figures 1 through 4 show subjects' responses to the questionnaire related to the use and disinfection of the PSP digital sensors during preclinical use. Due to multiple responses given by subjects for each question, the percentages may be higher than 100%. In Figure 1, subjects were asked what they liked most about using the PSP sensor. Twenty-five percent ($n=8$) indicated processing/XCP. film holding device used with the sensor, 21.9% ($n=7$) indicated the flexibility and thinness of the sensor, and 9.4% ($n=3$) stated that they liked to view the images on the computer. The remaining 12 subjects indicated that there was nothing they liked about using the PSP sensor.

In Figure 2, subjects were asked to state what they liked least about using the PSP sensors. Almost 60% (n=21) of respondents stated that, when placed inside the mouth, the sensor was bulky and uncomfortable for the patient. Another 36% (n=13) felt the infection control procedure was excessive, had too many steps, and took too much time. A smaller portion of the respondents did not feel comfortable with the sterility of the sensor (n=3) and felt that the sensors did not fit well on the film holding devices (n=8).

Subjects were then asked if they had any problems when performing the infection control protocol. The most frequent response, given by 40% (n=13) of the subjects, was that no problems occurred with following the infection control protocol, while 31% (n=10) stated that at one point the sensor touched the barrier envelope and/or gloves. A smaller group of respondents (n=7) commented that the infection control protocol they were assigned had too many steps to follow and, therefore, produced more work in preventing contamination of the sensor. The remaining seven subjects replied that they had a problem with re-inserting and sealing the sensor into the barrier envelope for fear that the sensor would fall on the countertop or create air pockets that could cause a problem when placing the sensor in the film holding device.

Lastly, in Figure 4, subjects were asked to describe how they perceived the infection control protocol used on the PSP sensor. Perceptions deviated into two major groups: 55% (n=18) of the subjects indicated the infection control protocol was not reliable in clinic, while 30% (n=10) felt the infection control protocol was effective in reducing contamination of the sensor. A smaller number of respondents felt the procedure was time consuming (n=6), and 3% (n=3) were unsure about the procedure and felt that more research was needed to determine if the infection control techniques used with the sensor were reliable in clinic. Subjects were also asked if they had any tears in the barrier envelope when removing the sensor from the oral cavity. Ninety-seven percent (n=32) stated they had no tears, while 3% (n=4) experienced tears.

Discussion

The transition from conventional film to intraoral digital sensors has created a new challenge to preventing cross-contamination in the dental setting. The purpose of this study was to test an infection control protocol for PSP digital sensors while simulating multiple uses in a preclinical setting.

In the study, when examining the PSP sensors for visual turbidity, discernable cloudiness was exhibited after 24 hours. Bacteria appeared to be present with nine of the 36 sensors (25%), but by 48 hours, 31 out of the 36 sensors were contaminated (86%). This result was validated by evaluating bacterial presence using optical densities. After incubation for 48 hours, 31 out of the 36 were contaminated (86%). Because optical density is not specific by bacterial type, the digital sensors were plated on MS agar to determine the presence of oral streptococci. The results of the plating concurred with the previous visual turbidity and optical density findings. By 24 hours, 61% of the sensor treated with the dry wipe and 100% of the sensors treated with the disinfectant wipe were contaminated with oral streptococci bacteria.

The results of this study create four issues for discussion. First, all the PSP sensors became contaminated with both infection control techniques after multiple uses. Two weeks prior to the study, subjects heard a lecture and observed a demonstration on how to remove sensors from the barrier envelope, but were not given the opportunity to practice the technique. Therefore, these inexperienced clinicians had never disinfected, torn, re-bagged, or placed the PSP sensor in a film holding device prior to participation in the study. The lack of experience resulted in four of the barrier-enveloped sensors not being sealed when they were removed from the cup for laboratory analysis. Because barrier envelopes were not sealed, infection control procedures may not have been followed correctly, which might explain the contamination found with the dry and disinfectant wipe. An alternative approach would be to calibrate the subjects before conducting the study to reduce the risk of error in infection control techniques used in the study. Despite this weakness in the study, the bacterial levels found in the study were low. Therefore, the barrier envelope proved to be an effective mechanism in reducing the amount of bacterial contamination on the PSP sensor. Even at 48 hours, the growth apparent on the sensors was low (10^8 CFU).

Another possible reason for the contamination of sensors with both infection control techniques may have resulted from perforations in the barrier envelope. In the questionnaire given to subjects one week following the study, subjects were asked if they had experienced any tears in the barrier envelope, and 5.6% indicated that they had. In contrast, in the Hubar et al. study, perforations occurred in 83% of the plastic sheaths.¹⁷ The difference in these findings between the studies

could have occurred because of the one week that lapsed between the preclinical experience and the questionnaire administration. Students' responses may not have accurately reflected what occurred in relation to the perforations.

A third point that can be summarized when examining the results of the study was that the dry-wipe group appeared to be more effective in preventing cross-contamination of the PSP digital sensor than the disinfectant-wipe group. Upon closer examination, these results may have occurred because the prophene was not used as a surface disinfectant (i.e. the disinfectant did not remain on the surface of the packet an adequate length of time to kill the bacteria). Prophene disinfectant, which is derived from phenol, is effective in killing bacteria on a variety of surfaces because it attacks and deteriorates the bacterial cell wall. For this to occur, the barrier envelope must initially be cleaned by spraying and wiping to reduce bioburden, then disinfected by reapplying the disinfectant and allowing the chemical to sit for at least 10 minutes on the contaminated surface to air dry.²¹

Regarding the study protocol, the disinfectant did not dry on the surface of the barrier envelope for 10 minutes. Instead, subjects immediately tore the barrier envelope after using the disinfectant as a cleaning agent. Because the surface of the barrier envelope may have still been wet, the barrier may have been more difficult to open, causing subjects to touch the sensor with contaminated gloved hands. In the questionnaire completed by each subject, almost 40% reported that the barrier envelope and gloves touched the digital sensor. Because residual saliva could have been left on the barrier envelope and/or gloves, cross-contamination of the sensor could have possibly occurred.

The results of the questionnaire concurred with Packota et al., who tested a one- and two-wipe method to disinfect conventional film.¹⁵ In the one-wipe method, test packets were wiped with a piece of sterile, moistened complex phenol solution followed by a dry paper towel, while the disinfectant was still wet on the surface. In the two-wipe method, the test packets were wiped with a dry paper towel to remove visible debris, then wiped again with the disinfectant and allowed to dry on the surface. Packota et al. discovered that the two-wipe method grew no bacteria, while the one-wipe method did. He stated that leaving the chemical disinfectant on the surface to air dry may eliminate any remaining organisms.¹⁵

Another explanation for the findings might be that, because the disinfectant was not given the opportunity to air dry, the wet solution on the surface of the film packet could have increased the spread of microbes and provided viable living conditions for the bacteria. Furthermore, the mixing of the disinfectant chemicals was not standardized. This may explain why the wet-wipe group was not effective in reducing microbial contamination. Differences in concentration of the disinfectant mixtures could have caused some solutions to be less potent than others. Also, the amount each subject sprayed on the paper towel to disinfect the barrier envelope was not standardized. To eliminate the variability in spraying a paper towel, a previous investigator submerged the packets into the disinfectant for approximately 60 seconds and then allowed them to air dry.⁴

Although the results of the study found contamination on both sensors, the amount of bacterial growth was minimal at baseline and 24 hours. If the disinfectant was not applied as a tuberculocidal agent, then one would have expected the bacterial contamination to have been prominent on all of the sensors, which was not indicated in the results. Sensors had >10⁸ bacterial growth only after sensors were amplified during incubation.

The results of this study suggest that radiographers should re-evaluate their infection control protocol for the use of intraoral digital sensors. Considerations should be given to the use of a dry-wipe over a wet-wipe procedure. Although sensors became contaminated with both infection control techniques, the bacterial load was less in the dry-wipe group after 24 hours of incubation. These findings were supported by the questionnaire data that revealed that half of the subjects believed the infection control procedures were unreliable. A questionnaire response revealed that subjects felt that the wet-wipe required too many steps and believed steps could be easily skipped or not followed correctly, causing cross-contamination to occur. Limitations to the study can be related to the study population. Although subjects were randomized into two test groups, the population was not stratified by mean average of radiographic competencies, prior dental assisting experience, or male to female ratios. Another possible weakness to the study was that more subjects were present on day one of the study than were on day two. Furthermore, due to the lack of XCP instruments, half of the subjects used the Stabe) for the study, resulting in differences between both study days. The results of this study are confined only to this study population.

Conclusion

The effectiveness of two infection control techniques was tested with cross-contamination of the PSP digital sensor occurring with both techniques. Though contamination was evident, the amount of bacterial contaminants was low. However, it appeared that the dry-wipe method was more effective in reducing the risk of bacterial contamination of the PSP sensor when compared to the disinfectant wipe. The question still remains as to whether barrier-enveloped sensors treated effectively with a surface disinfectant will eliminate cross-contamination with multiple uses of the digital sensors. Additional research needs to be conducted to evaluate the most effective infection control procedure and to develop an appropriate protocol for use of PSP sensors.

Acknowledgements

Notes

Correspondence to: Wendy Negron at wnegron@omniipharma.com

References

1. Geist JR, Stefanac SJ, Gander DL. Infection control procedures in intraoral radiology: a survey of Michigan dental offices. *Clin Prev Dent.* 1990;12(2): 4-8.
2. Kelly W. Radiographic asepsis in endodontic practice. *Gen Dent.* 1989;37(4): 302-312.
3. Neaverth E, Pantera EA. Chairside disinfection of radiographs. *Oral Surg Oral Med Oral Pathol.* 1991;71(1): 116-9.
4. Parks ET, Farman AG. Infection control for dental radiographic procedures in U.S. dental hygiene programmes. *Dentomaxillofac Radiol.* 1992;21(1): 16-20.
5. Stanczyk D, Broome JC. Microbiologic contamination during dental radiographic film processing. *Oral Surg Oral Med Oral Pathology.* 1993;76: 112-9.
6. Tullner JB, Zeller G, Hartwell G, Burton J. A practical barrier technique for infection control in dental radiology. *Compendium.* 1992;13(11): 1054-6.
7. Wolfgang L. Analysis of a new barrier infection control system for dental radiographic film. *Compendium.* 1992;13(1): 68-71.
8. Wyche C. Infection control protocols for exposing and processing radiographs. *J Dent Hyg.* 1996;70(3): 122-6.
9. Hubar JS, Oeschger MP, Reiter LT. Effectiveness of radiographic film barrier envelopes. *Gen Dent.* 1994;42(5): 406-8.
10. Hubar JS, Oeschger OP. Optimizing efficiency of radiograph disinfection. *Gen Dent.* 1995;43(4): 360-2.
11. White SC, Glaze S. Interpatient microbiological cross-contamination after dental radiographic examination. *J Am Dent Assoc.* 1978. May;96: 801-4.
12. Glass BJ. Infection control in dental radiology. *N Y State Dent J.* 1994;60(4): 42-5.
13. Autio KL, Rosen S, Reynolds NJ, Bright JS. Studies on cross-contamination in the dental clinic. *J Am Dent Assoc.* 1980. March;100: 358-361.
14. Bartoloni J, Charlton D, Flint D. Infection control practices in dental radiology. *Gen Dent.* 2003;51(3): 264-271.
15. Packota GV, Komiyama K, Saskatoon S. Surface disinfection of saliva-contaminated dental x-ray film packets. *J Can Dent Assoc.* 1992;58(9): 747-751.
16. Hokett SD, Honey JR, Ruiz F, Baisden K, Hoen MM. Assessing the effectiveness of direct digital radiography barrier sheaths and finger cots. *J Am Dent Assoc.* 2000;131(4): 463-7.
17. Hubar JS, Gardiner DM. Infection control procedures used in conjunction with computed dental radiography. *Int J Comput Dent.* 2000;3(4): 259-267.
18. Wenzel A, Frandsen E, Hintz H. Patient discomfort and cross-infection in bitewing examination with a storage phosphate plate and a CCD-based sensor. *J Dent.* 1998. Sept;27: 243-6.
19. Koch S, Wagner IV, Schneider W. Effective and quality-controlled use of digital radiography in dental practice. *Int J Comput Dent.* 2000;3: 107-118.
20. Peterson CA, Mauriello SM, Arnold RR, Webster-Cyrique J. Infection control for a digital imaging sensor. *J Dent Res.* 2002;81(Special Issue A): A-111.
21. Organization for Safety and Asepsis Procedures . Infection control in practice: dentistry's resource for infection control and safety. 2002. Aug;1(3).