Long-term Bone Response to Titanium Implants Coated with Thin Radiofrequent Magnetron-Sputtered Hydroxyapatite in Rabbits

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Purpose: *The present study was designed to investigate the long-term bone response around machined screw-type uncoated and calcium phosphate (CaP) –coated commercially pure titanium implants.* ***Materials and Methods:*** *Using a magnetron sputtering technique, implants with a CaP coating similar in composition and CaP ratio to hydroxyapatite were produced. Heat treatment was subsequently used to increase the crystallinity of the coatings. Four types of coatings (0.1 and 2.0 μm amorphous and 0.1 and 2.0 μm crystalline) were manufactured; uncoated implants served as a control. Three hundred twenty implants (64 of each type) were randomly placed in the tibial cortical and trabecular femoral bones of 40 rabbits. The rabbits were sacrificed 9 months after implant placement.* ***Results:*** *Histomorphometric evaluation carried out on ground sections revealed that the crystalline CaP coatings achieved the highest bone-implant contact in both tibiae and femora compared with amorphous CaP-coated and uncoated titanium.* ***Discussion:*** *The present study suggests that submicron crystalline hydroxyapatite coating adds bioactive properties to titanium oral implants.* ***Conclusion:*** *An ultra-thin, 0.1-μm crystalline CaP coating can elicit and maintain an improved long-term bone response compared to amorphous coated or uncoated Ti implants, without any adverse tissue reactions.* (More than 50 references.) INT J ORAL MAXILLOFAC IMPLANTS 2004;19:498–509

Key words: *bone, dental implants, histomorphometry, hydroxyapatite, magnetron sputtering, titanium*

Osseointegrated titanium implants have been used in dentistry with good results for more than 3 decades.[1] Nonetheless, there is room for improvement. To improve the process of osseointegration, a number of surface modifications have been proposed. It has not been possible to establish the clinical superiority of any particular surface modification[2]; in particular, no reliable clinical trials have evaluated calcium phosphate (CaP) coatings on dental implants.[2] Several experimental observations[3–5] have suggested that the process of bone integration may occur faster around implants coated with CaP, and this could be of clinical importance.

In a retrospective controlled clinical trial,[6] dental implants coated with hydroxyapatite (HA) were compared with titanium implants in 166 patients. After following the implants for up to 6 years, the authors concluded that the performance of HA-coated implants was similar to that of titanium implants. In another retrospective controlled clinical trial,[7] 313 HA-coated implants and 889 titanium plasma-sprayed implants were placed in 479 patients. A higher initial survival rate was observed for HA-coated implants, but after 4 years the inverse relationship was found. Most late failures were reported to be associated with inflammatory changes involving HA-coated implants (ie, peri-implantitis).

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Survival rates of 79% for HA-coated implants and 97% for titanium implants were reported after up to 8 years of follow-up.

In orthopedics, both short-term[8,9] and long-term[10] clinical trials on HA-coated hip prostheses have been published. In a controlled clinical trial,[11] the performance of uncemented HA-coated proximal porous femoral stem hips was compared to that of porous-coated ones in 15 patients. After 1 year, no major differences were observed. Furthermore, in a randomized clinical trial[12] comparing uncemented cobalt-chrome plasma-sprayed porous-coated and HA-coated arthroplasties in 78 patients, no difference in survival was found between 2 types of surfaces after 8 years. In a 5-year randomized controlled clinical trial comparing uncemented plasma-spayed HA-coated total knee arthroplasties with uncemented porous-coated titanium ones, no differences in the clinical outcome were found.[13]

A variety of techniques for manufacturing CaP coatings have been developed, such as plasma spraying,[14] sputtering,[15,16] and pulsed laser deposition.[17] Since the various techniques involve different physical and chemical processes, it is clear that the coating properties are dependent upon the technique used. It is possible that the use of coatings with different properties can result in differences in the biologic response. Furthermore, the biologic response of peri-implant tissue might vary in different types of tissue (eg, cortical bone, trabecular bone).[18] Therefore, it is difficult to compare results from different studies.

Several studies have reported on plasma-sprayed CaP surfaces.[14,19–21] Plasma spraying is the most frequently used technique for applying commercial CaP coatings to medical devices. Plasma-sprayed surfaces usually have irregular surface topographies[22] and consist of a mixture of amorphous and crystalline CaP phases of varying chemical compositions.[20,23–26] Different observations of the biologic response to such surfaces have been reported.[3,21,23,27] Some studies have shown that HA plasma-sprayed coatings tend to dissolve in vivo.[3,27,28]

The sputtering technique allows for the deposition of very thin, stable, homogenous coatings on metal substrates.[29,30] However, the stability of the coating is dependent on the degree of crystallinity; crystalline coatings are more stable than amorphous ones.[29,31,32]

The biologic response to CaP coatings produced with sputtering techniques has also been investigated extensively.[18,33–36] The early biologic response to CaP coatings has been investigated in short-term experimental trials,[5,36] but long-term data are scarce and present contradictory results,[4,19] probably because different types and characteristics of CaP coatings were investigated.

The aim of the present study was to investigate the long-term bone response to micron and submicron layers of magnetron-sputtered HA as a function of surface crystallinity in 2 different implantation sites.

MATERIALS AND METHODS

Implants

Four different types of CaP coating were used; uncoated titanium implants (the controls) were also tested. All implants were machined from commercially pure titanium rods (ASTM grade 1). The surface topography of the implants was similar to the topography of commercial implants manufactured by Nobel Biocare, Göteborg, Sweden. The implants, which were 4 mm long and 3.75 mm wide, had threads down the entire length of the cylinder. A threaded hole inside the implant along the implant axis allowed for mounting during handling and the various surgical procedures.

All implants were thoroughly cleaned with buthanol followed by ethanol using ultrasonic equipment. Implants to be coated were mounted in a radiofrequency (RF) magnetron sputtering apparatus with a base pressure of 10^{-6} mb. A maximum of 40 implants were processed during each sputtering procedure. The sputtering target was HA (HC CAM, Leiden, The Netherlands). Sputtering was performed in a mixture of argon and reactive gases to obtain the desired HA stoichiometry. Implants were designated by lottery to 1 of the following groups:

- Heat-treated, 0.1 μm thick, crystalline (group A)
- Heat-treated, 2.0 μm thick, crystalline (group B)
- Non–heat-treated, 0.1 μm thick, amorphous (group C)
- Uncoated machined titanium (control) (group D)
- Non–heat-treated, 2.0 μm thick, amorphous (group E)

Heat treatment of the coatings was carried out in a specially designed gas flow reactor at 873 K for 14 hours in a saturated mixture of water vapor and synthetic air.

Cleaning and Sterilization

All implants were exposed to UV-ozone for 30 minutes. The implants were mounted by pairs in cleaned titanium containers, which were sealed with 2 tight-fitting lids. All implants were sterilized with γ-radiation; a minimum dose of 25 kGy was used.

Five percent of the implants, randomly selected from different batches, were tested for the presence of microorganisms and bacterial endotoxins on their surfaces. The implants were immersed in a fluid thioglycolate medium (Difco Laboratories, Detroit, MI) and incubated at 37°C for 3 weeks to confirm their sterility. The amount of bacterial endotoxin was estimated by applying the Limulus Amebocyte Lysate (LAL) test. The LAL test is a standardized, quantitative chromogenic method used at the Department of Clinical Bacteriology, Sahlgrenska Academy at Göteborg University. Sterility was validated with respect to colony-forming units (CFUs) and endotoxins.

Material Surface Characterization

The coating thickness and topography of the surfaces were measured with a stylus profilometer. The stoichiometry of the coatings was analyzed by Fourier transform infrared spectroscopy (FTIR) and inductively coupled plasma optical emission spectroscopy (ICP-OES). FTIR was performed using a Mattson Cygnus 100 with an attenuated total reflectance (ATR) attachment. The CaP coating was removed from 5 different implants from 4 different production lots with 10 mmol/L nitric acid. The coating crystallinity was examined with x-ray diffraction (XRD) in a Siemens D 5000 diffractometer (Siemens, Munich, Germany). The XRD and FTIR measurements were performed on 2.0-μm thick coatings.

Animals and Surgical Procedures

According to a randomized implant placement plan, 320 implants (64 of each type) were placed in 40 adult female New Zealand White rabbits, which weighed 3 to 6 kg (mean 5 kg) and were fed ad libitum.

Prior to surgery, the animals were anesthetized by intramuscular injections of fluanisone (0.7 mg/kg body weight Hypnorm; Janssen, Brussels, Belgium) and an intraperitoneal injection of diazepam (1.5 mg/kg body weight Stesolid; Dumex, Copenhagen, Denmark). Additional fluanisone was given as needed during surgery (approximately every 25 minutes). The limbs were shaved and disinfected with chlorhexidine (5 mg/mL; Pharmacia, Stockholm, Sweden). The operations were performed under sterile conditions and were designed to cause minimal trauma to the tissues.

Each animal received 4 implants of the same type in one leg and 4 implants of another type in the contralateral leg. Two implants were placed in each tibial metaphysis (proximally and distally) and 2 implants in each femoral condyle (laterally and medially). The implant placement areas were exposed separately through skin incisions and blunt dissection of the underlying tissues, including the periosteum. In the tibial metaphysis, 2 holes were drilled 1 cm apart with a dental guide drill at 2,500 rpm using a titanium template. One hole was drilled in the lateral femoral condyle and 1 in the medial femoral condyle. The holes were sequentially enlarged using larger drills (a 2-mm-wide twist drill, a pilot drill, and a 3-mm-wide twist drill) under profuse saline irrigation. The holes were tapped with a screw tap at 16 rpm under saline irrigation. Implants were removed from the titanium cylinders using a titanium forceps and placed gently into the holes using a titanium screwdriver so that the upper surface of the implant was level with the cortical bone surface. The operation site was then rinsed with saline, and the tissues were sutured in separate layers with Vicryl 5-0 (Ethicon, Somerville, NJ) and finally with Suturamid 3-0 (Ethicon). The mean surgery time was about 2 hours per animal.

For 7 days postoperatively, animals were injected intramuscularly with enrofloxacin (vet 25 mg/mL, 1 mL/day Baytril; Bayer HealthCare, Leverkusen, Germany) and analgesics (0.05 mg/kg/day Buprenorphine; 0.05 mg/kg/day Temgesic; Reckitt and Colman Pharmaceutical, Richmond, VA). The animals were housed separately during this period. They were then moved to group housing and observed until 9 months after surgery.

Specimen Processing and Analysis

Animals were sacrificed by an intravenous overdose of pentobarbital (Mebumal, ACO Läkemedel, Solna, Sweden) and fixed by perfusion with 2.5% glutaraldehyde in 0.05 mol/L cacodylate buffer, pH 7.4, via the left ventricle of the heart. Implants and their surrounding tissues were removed en bloc, immersed in glutaraldehyde for 1 to 2 weeks, and postfixed in 1% osmium tetroxide for 2 hours. After dehydration in a graded series of ethanols, the specimens were embedded in plastic resin (LR White, The London Resin Company, Hampshire, UK) and divided through the long axis of each implant using a band saw with a diamond blade (Exakt Apparatebau, Norderstedt, Germany). One half of each specimen was used to prepare an approximately 100-μm-thick section, which was ground to a thickness of 15 μm or less (DPU-3; Struers Scientific Instruments, Copenhagen, Denmark).[37] The specimens were stained with 1% toluidine blue.

Qualitative evaluation, morphometric measurements,[38–40] and photographs were made using a Leitz Microvid Morphometric System equipped with a personal computer and dedicated software (MicroMacro, Göteborg, Sweden) (Fig 1). Measurements

were carried out directly in the microscope. The morphometric measurements were performed blindly by the first author using 10× magnification. For each implant, the mean percentages of bone-implant contact and bone area in each thread (left and right side) and in 5 consecutive threads together were calculated. In addition, the mean percentage of bone-implant contact and bone area in the distal threads (4 and 5) of all implants were calculated and compared.

Statistical Analysis

The statistical analysis was performed at the Department of Statistics, Göteborg University, using a balanced incomplete randomized block design. The analysis of variance (ANOVA) model was conducted with a general linear model (GLM) procedure using a statistical program package (SPSS, Chicago, IL), with adjustment for multiple comparisons. Results of the analyses are presented in Tables 1 and 2. The presented model based on estimated means differs slightly from the means calculated from the individual implants because of the incomplete design. The estimated means are more accurate, as they take into account that the means were calculated using a different subsample for every implant type. The standard error of the mean (SEM) is based on the common model variance, mean square error (MSE), also adjusted according to the incomplete design of the model.

Three levels of significance were used: $P < .05$ (*), $P < .01$ (**), and $P < .001$ (***).

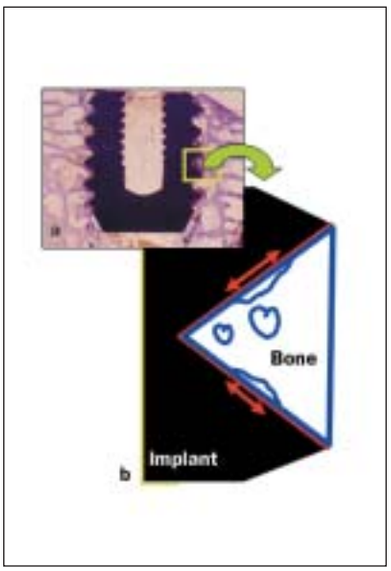


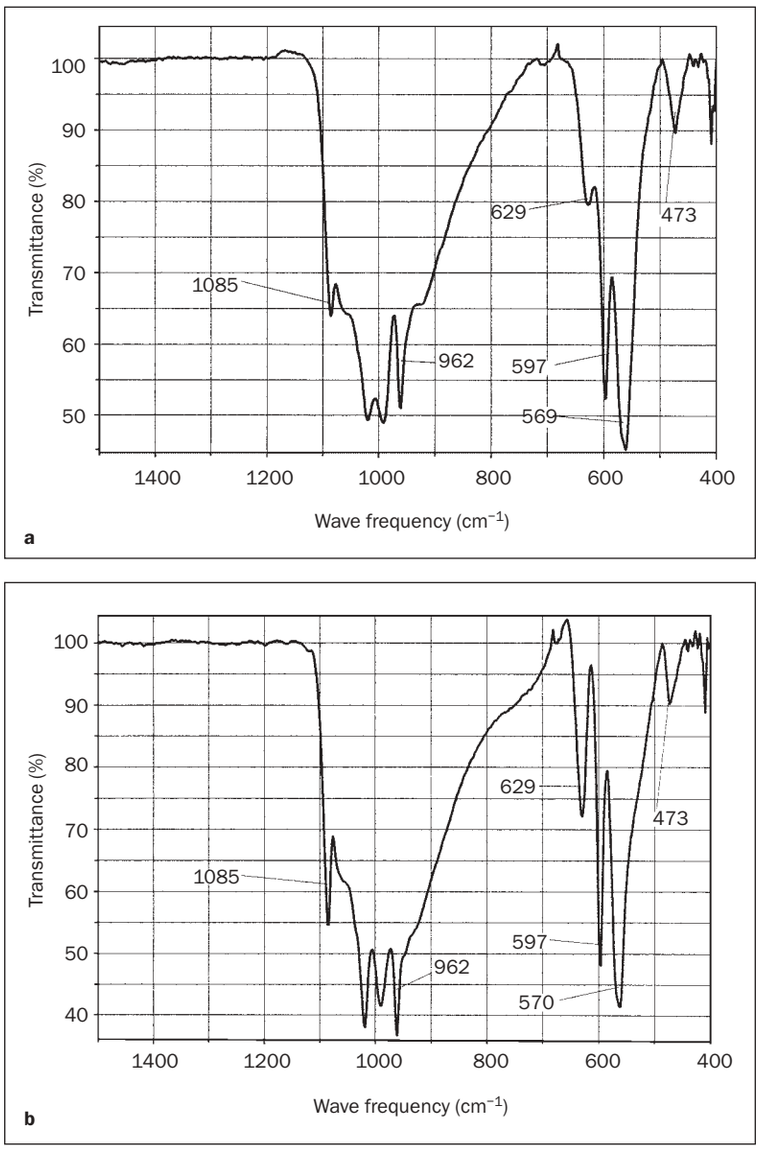
Fig 1 *(a)* A histologic specimen showing the hole along the implant axis used for mounting. *(b)* A schematic illustration of the morphometric measurements of bone-implant contact and bone area within a thread. The percentage of bone-implant contact was calculated after measuring the implant perimeter *(red line)* and the perimeter with bone contact. Red arrows indicate areas not showing bone-implant contact. The percentage of bone area within a thread was calculated by measuring the total area within the thread occupied by tissue *(blue triangle)* and the area occupied by bone. White areas surrounded by a blue line indicate soft tissues.

Table 1 Bone-Implant Contact and Bone Area in the Tibia After 9 Months

	Bone contact				Bone area			
Implant	Mean % threads 1 to 5	SEM	Mean % threads 4 and 5	SEM	Mean % threads 1 to 5	SEM	Mean % threads 4 and 5	SEM
A	55.45		47.48		63.88		34.22	
B	52.61		50.56		60.01		31.47	
C	36.16	1.93	32.57	2.72	68.70	2.46	52.30	5.37
D	46.06		43.43		71.78		54.39	
E	45.00		47.72		72.41		58.60	

Table 2 Bone-Implant Contact and Bone Area in the Femur After 9 Months

	Bone contact				Bone area			
Implant	Mean % threads 1 to 5	SEM	Mean % threads 4 and 5	SEM	Mean % threads 1 to 5	SEM	Mean % threads 4 and 5	SEM
A	70.66		66.69		64.18		48.12	
B	72.07		68.09		55.05		37.69	
C	53.35	2.69	51.28	3.23	82.45	3.17	75.14	5.38
D	55.50		54.77		82.36		75.66	
E	55.59		54.62		80.14		73.47	



Figs 2a and 2b FTIR spectra from *(a)* non–heat-treated and *(b)* heat-treated CaP-coated titanium. The indicated adsorption bands at 630 and 600 cm^{-1} corresponded to OH lattice and PO_4-v4 internal vibration modes, respectively.[42] The pronounced intensity of the OH band at 630 cm^{-1} in *b* corresponded very well to calcium hydroxyapatite ($Ca_{10}[PO_4]_6[OH]_2$).[43,44] The OH band at 630 cm^{-1} in *a* is less pronounced.

RESULTS

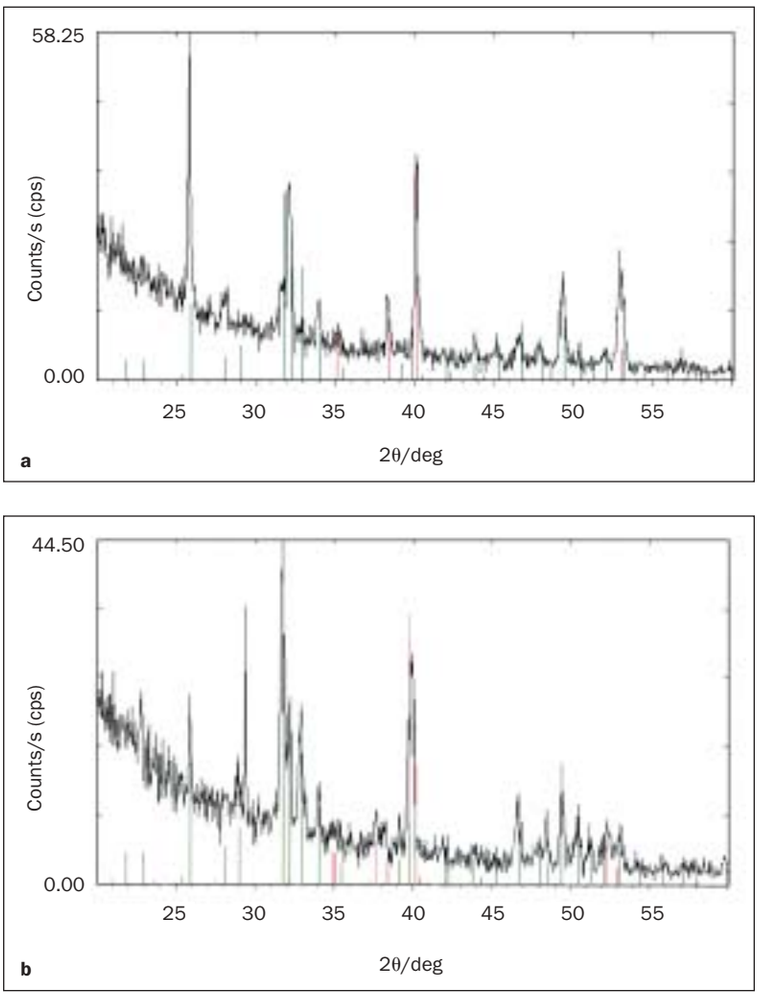
Sterilization

Microbiologic tests showed that the concentrations of CFUs and endotoxins were less than 1 CFU/implant and less than 0.01 endotoxin unit/implant.

Material Surface Characterization

The average coating thicknesses, measured with a stylus profilometer, were 0.1 ± 0.01 μm and 2.0 ± 0.1 μm. Stylus profilometric analysis demonstrated a comparable surface topography for all implants. ICP-OES demonstrated a Ca/P molar mean ratio of 1.665 (SD 0.018). The FTIR spectra (Figs 2a and 2b) corresponded very well to those of calcium hydroxyapatite.[41–43] The pronounced intensity of the OH lattice vibration mode at 630 cm^{-1} for the heat-treated samples indicated that the coating was $Ca_{10}(PO_4)_6(OH)_2$. The intensity of this mode for non–heat-treated samples was less pronounced (Fig 2a). However, all the peaks appeared to be broadened, indicating that the chemical environment for each of the groups represented in the spectra was not as uniform as for the heated samples. The most probable reason is the large difference in the crystallinity of the heat-treated specimens compared to the non–heat-treated ones. The latter consisted of much smaller crystallites, resulting in a substantial broadening of the FTIR peaks. This conclusion was supported by the XRD measurements.

The XRD spectra (Figs 3a and 3b) of heat-treated CaP coating revealed a dramatic increase in



Figs 3a and 3b XRD spectra from *(a)* non–heat-treated and *(b)* heat-treated CaP-coated titanium. In *a,* a few peaks can be seen in the spectra of the non–heat-treated sample, which could be related to the CaP surface. The surface was therefore not completely amorphous. The position of the peaks corresponded well with some peaks of the standard for HA (Joint Committee on Powder Diffraction Standard 9-432), *(green bars)* while the measured intensities of the peaks diverged strongly from the standard. Titanium *(red bars)* could also be identified. In *b,* HA can clearly be identified. A comparison of this sample with the standard for HA gave a good correspondence of the peaks considering both the positions and the intensities. Both titanium and titanium dioxide *(pink bars)* could be identified.

crystallite sizes compared to non–heat-treated samples. The heat treatment process resulted in increased crystallite size as well as a gradual increase of the coating crystallinity. The XRD pattern was in agreement with the HA standard of the Joint Committee on Powder Diffraction (standard 9-432).

Histologic Evaluation

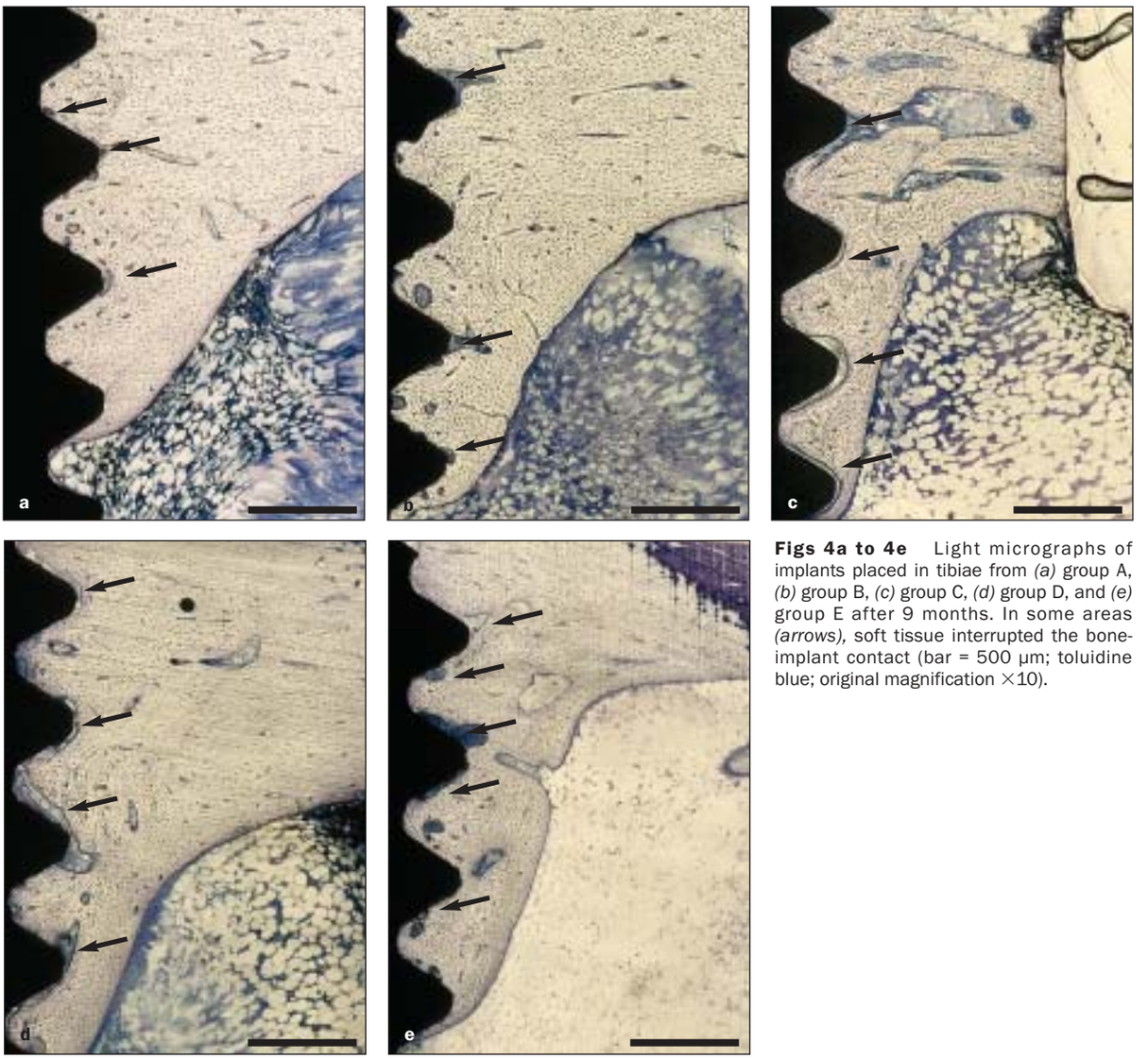
Tibia. Mature, well-organized bone filled most of the proximal threads (1 to 3) of all implant types (Figs 4a to 4e). Intimate bone-implant contact was established, although it was occasionally interrupted by soft tissue.

Femur. Implants were surrounded by mature bone within the threads; mature bone was also in direct contact with the implant surface (Figs 5a to 5e). A clear difference could be observed between implants with a crystalline coating and implants with an amorphous coating or no coating. Crystalline implants had a thin bone collar in contact with their surfaces, whereas the areas within the threads were not occupied completely by bone (Figs 5a and 5b). Conversely, bone filled almost the entire area within the threads of the amorphous coated and uncoated implants (Figs 5c to 5e).

No signs of inflammatory reaction could be detected in the tissues surrounding the implants.

Histomorphometry

Data for bone-implant contact and bone area in tibiae and femora are presented in Tables 1 and 2 and in Figs 6 and 7.



Figs 4a to 4e Light micrographs of implants placed in tibiae from *(a)* group A, *(b)* group B, *(c)* group C, *(d)* group D, and *(e)* group E after 9 months. In some areas *(arrows),* soft tissue interrupted the bone-implant contact (bar = 500 μm; toluidine blue; original magnification ×10).

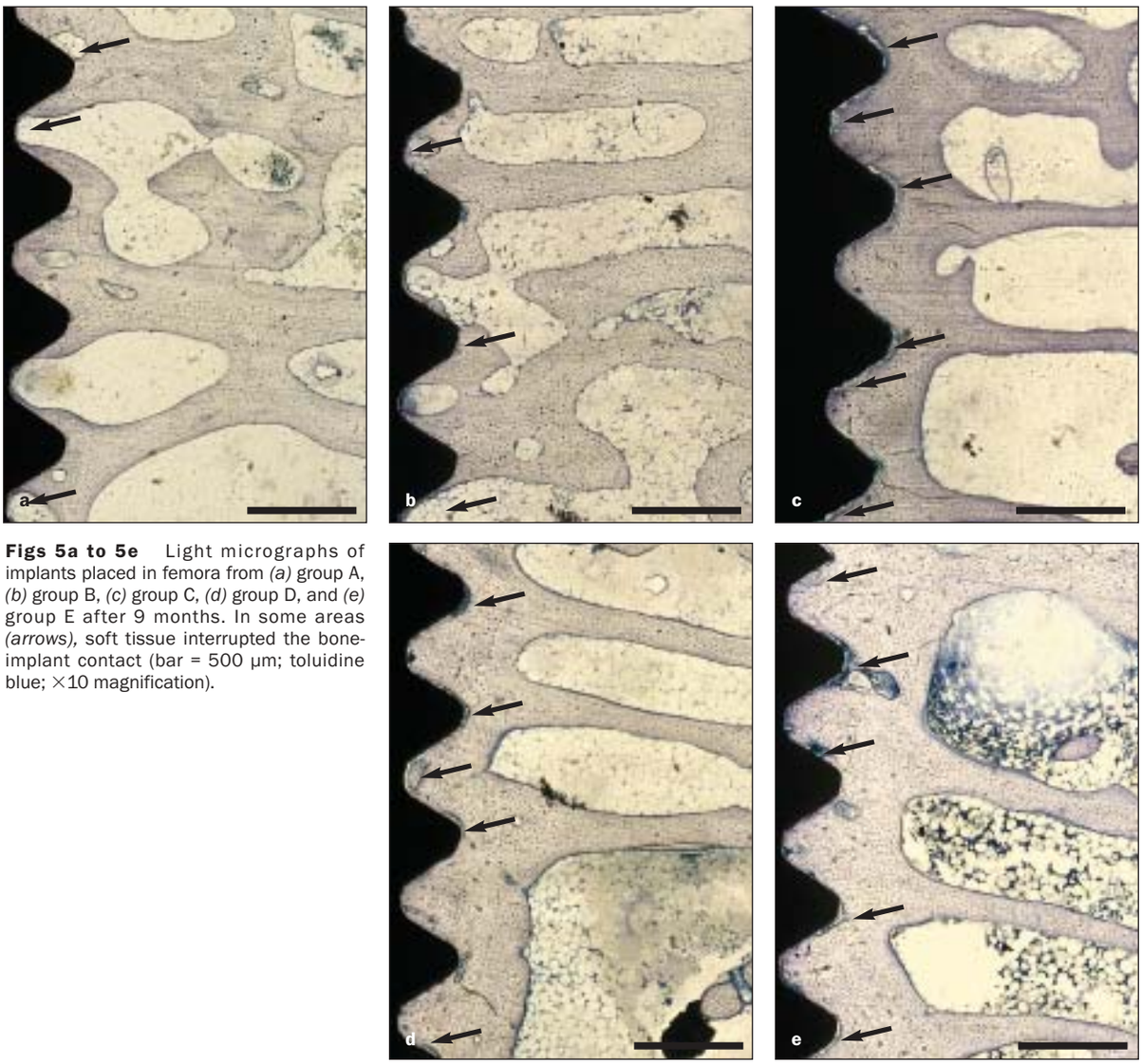
After 9 months in the tibia, implant A had significantly higher bone-implant contact than implants C ($P < .001$), D ($P < .05$), or E ($P < .01$) in threads 1 to 5. Implant B had significantly higher bone-implant contact than implant C ($P < .001$). In addition, implants D and E showed significantly higher bone-implant contact than implant C ($P < .01$ and $P < .05$, respectively). No significant differences were observed between implants D and E (Table 1; Fig 6a).

When considering threads 4 and 5, there was significantly higher bone-implant contact for implants A and B compared to implant C ($P < .01$ and $P < .001$, respectively). In this respect, implant E had a significantly higher bone-implant contact than implant C (Table 1; Fig 6a) ($P < .01$).

Implants D and E had significantly more bone area in threads 1 to 5 than implant B ($P < .05$ and $P < .01$, respectively) (Table 1; Fig 6b). With respect to threads 4 and 5, implant E had significantly more bone in these threads than implant A ($P < .05$) or B ($P < .01$). Implant D had significantly more bone than implant B ($P < .05$) (Table 1 and Fig 6b).

In femora, implant A had significantly more bone-implant contact than implants C ($P < .001$), D ($P < .01$), and E ($P < .01$). Implant B also had significantly more bone-implant contact than implants C ($P < .001$), D ($P < .001$), and E ($P < .01$). In regard to threads 4 and 5, implant A had significantly more bone-implant contact than implant C ($P < .05$), and implant B had significantly more than implants C ($P < .01$), D ($P < .05$), or E ($P < .01$) (Table 2 and Fig 7a).

Implants C, D, and E had significantly more bone area than implants A ($P < .01$) and B ($P <$



Figs 5a to 5e Light micrographs of implants placed in femora from *(a)* group A, *(b)* group B, *(c)* group C, *(d)* group D, and *(e)* group E after 9 months. In some areas *(arrows),* soft tissue interrupted the bone-implant contact (bar = 500 µm; toluidine blue; ×10 magnification).

.001) (Table 2, Fig 7b). Implants C, D, and E also had significantly more bone area than implants A ($P < .01$ for C and D; $P < .05$ for E) and B ($P < .001$) in regard to threads 4 and 5 (Table 2, Fig 7b).

DISCUSSION

The aim of the present study was to investigate the long-term performance of RF magnetron-sputtered coatings in rabbit tibiae and femora. Machined screw-type CaP-coated titanium implants with 4 types of coatings as well as uncoated Ti implants were placed in cortical and trabecular bone in rabbits. Histomorphometric evaluation showed that more bone was in contact with implants with crystalline coating compared with implants with amorphous coating and uncoated implants in both implantation sites. These findings confirm earlier short-term observations.[5] After 9 months, crystalline coatings maintained the intimate bone-implant contact that was established after 6 weeks without any sign of inflammatory reaction in the tissues around the implants.[5]

Several long-term animal studies on the biologic behavior of HA coatings have been published.[4,19,45,46] It has been shown that plasma-sprayed HA coatings induced significantly more bone-implant contact than titanium after 6 to 12 months.[19,45,46] The plasma-sprayed coatings showed signs of resorption in all studies, but only in 1 study[19] were inflammatory cells seen around HA-coated implants. In another animal experiment, implants with electrophoretically applied

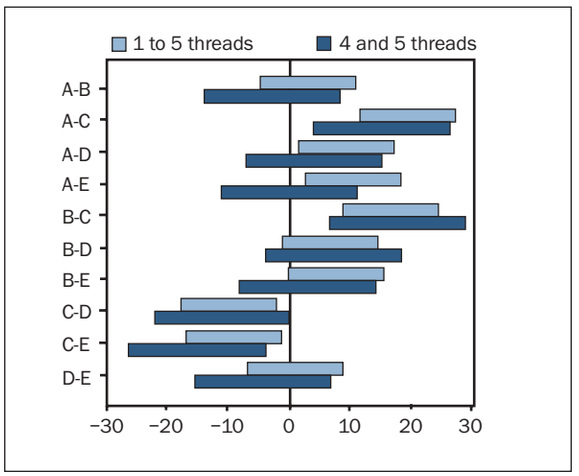


Fig 6a Statistical comparison of bone-implant contact in tibiae for pairs of implants. Confidence intervals of implant differences are shown as horizontal bars. The following significant differences between implants were found in regard to threads 1 to 5: A > C***, A > D*, A > E**, B > C***, D > C**, E > C*. For threads 4 and 5, A > C**, B > C***, E > C**.

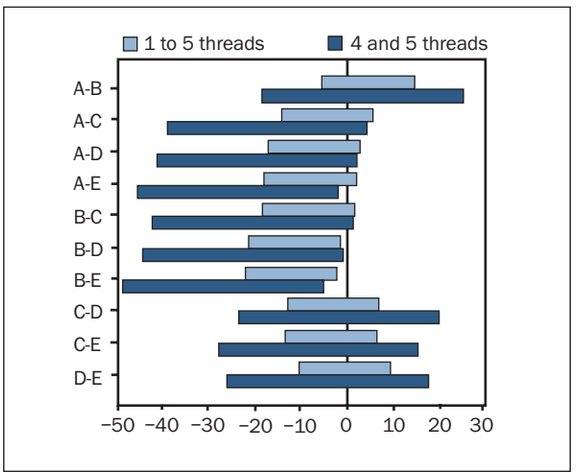


Fig 6b Statistical comparison of bone area contact in tibiae for pairs of implants. Confidence intervals of implant differences are shown as horizontal bars. Confidence intervals show significant differences at the 5% level if the whole interval lies either to the right or the left of the zero line. The intervals are based on exactly the same conditions as the corresponding hypothesis tests.[44] Two statistically significant differences between implants were found in regard to threads 1 to 5: D > B* and E > B**. For threads 4 and 5, D > B*, E > A*, and E > B**.

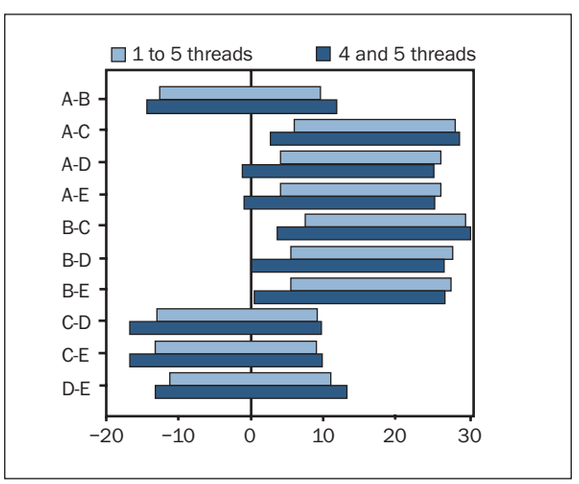


Fig 7a The morphometric data for bone-implant contact in femora are presented as horizontal bars when pairs of implants were compared with one another. The following significant differences were for bone-implant contact between implants in regard to threads 1 to 5: A > C***, A > D**, A > E**, B > C***, B > D***, and B > E**. For threads 4 and 5, A > C*, B > C**, B > D*, and B > E*.

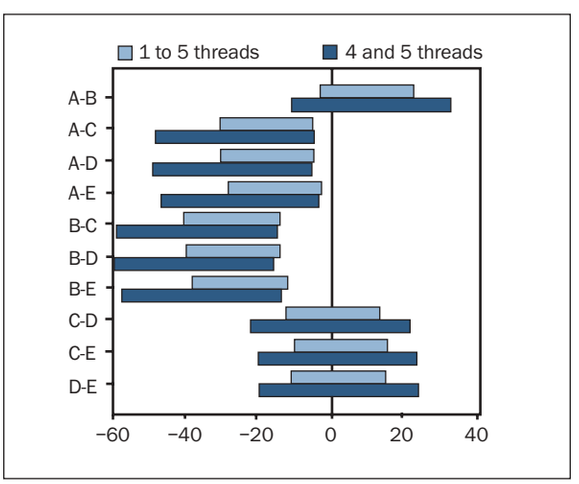


Fig 7b The morphometric data for bone area in femora are presented as horizontal bars when pairs of implants were compared with one another. They show significant differences at the 5% level if the whole bar lies either to the right or the left of the zero line. The intervals are based on exactly the same conditions as the corresponding hypothesis tests.[44] The following significant differences were for bone area between implants in regard to threads 1 to 5: C > A**, C > B***, D > A**, D > B***, E > A**, E > B***. For threads 4 and 5, C > A**, C > B***, D > A**, C > B***, E > A*, and E > B***.

CaP coatings were compared with uncoated titanium implants for 6 months.[4] No significant difference in bone-implant contact was found between HA-coated and uncoated implants. In addition, inflamed cells and resorption of the coatings were reported.

Amorphous HA coatings dissolve within 4 weeks[31]; indeed, the observed bone response to the amorphous coatings in the present study was similar to that observed around uncoated implants. It was not possible to detect the micron and submicron

coatings at the light microscopic level. From SEM analysis, it has been reported that 0.1-μm thick coatings are not visible, but 1-μm thick coatings have been seen occasionally.[47] Degradation of the CaP coatings has been suggested to be caused by inflammatory cells[48] or low pH level during the initial wound healing.[49] However, no harmful coating fragments were seen around the coated implants in the present study.

There have been studies demonstrating that a higher degree of crystallinity of the CaP ceramics may resist dissolution of the coating.[28] Furthermore, the significance of coating dissolution and the ability of the dissolved coating to form CO_3-apatite crystals as foci for enhanced bone formation is often discussed in the literature.[31,36,50] On the other hand, apart from the composition characteristics of the CaP coatings, the topography of various surfaces has been shown to be important for the biologic outcome.[51] Therefore, the possibility that the chemical and microtopographic properties had synergistic effects cannot be excluded.[20] In an in vitro investigation,[52] increased matrix production on a rough HA surface was reported. Previous short-term data have shown that a crystalline coating is a prerequisite for improved early bone response.[5] Provided that the amorphous coatings were dissolved within a few weeks, the data indicated that the role of calcium ion release could be ignored. Whether the animal model used had any impact on behavior of the CaP coatings is unknown. However, it has been suggested that certain experimental animals, including rabbits, are fast healing.[36]

In another study,[27] cylindric Ti-6Al-4V implants, each coated with a 50-μm-thick layer of either plasma-sprayed HA, alpha-tricalcium phosphate, or tetracalcium phosphate, were compared with noncoated implants after 3, 5, 15, and 28 months in dog femora. HA coatings showed shear strength of 30 MPa after 3 months of implantation, which did not increase significantly after 28 months of implantation. After 5 months, titanium showed the highest shear strength (21 MPa). HA coatings showed significant coating degradation after 5 months. Multinuclear cells, osteoclasts, and osteoblasts were seen at the interface of coated implants with degraded coatings. In another study,[53] 75-μm-thick HA-coated Ti-6Al-4V and bead-blasted commercially pure titanium implants were placed in dog femora for 8 months. Interface shear strength and stiffness evaluation revealed significantly higher values for HA-coated implants. Significant interface strength was more rapidly established around HA-coated implants (5 weeks) than bead-blasted titanium and was better maintained over time. No statistically significant change in the coating thickness up to 8 months could be observed.

In the present study no biomechanical testing of the implant osseointegration (torque, pull/pushout tests) was done. However, a correlation between biomechanical tests and amount of bone in contact with the implant has been demonstrated.[54]

CONCLUSIONS

The presented data showed that the crystalline coatings were superior to amorphous and uncoated implants with respect to bone-implant contact. No additional advantage was observed when a thicker crystalline coating (2.0 μm) was applied. In fact, there was little difference between the results obtained with amorphous-HA-coated implants and those obtained with uncoated implants. The long-term data presented in this study are in agreement with previous findings after 6 weeks.[5] Different types of bone (cortical and trabecular) react differently to different implant materials. Furthermore, the amount of bone and the bone-implant contact values around implants increased with time.

Based on the present results, it was concluded that 100-nm-thick crystalline RF magnetron sputtered coatings provided a better long-term bone response compared with amorphous and noncoated implants. The positive long-term effect of the nanometer thick crystalline coatings appeared to be governed by the early tissue-biomaterial interactions. Taken together, the data suggest that a 0.1-μm-thick crystalline HA coating could be of clinical interest, because of the following advantageous properties: more rapid bone formation,[5] higher degree of bone-implant contact maintained, and the absence of coat-fragment–induced inflammation.

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