

Antibacterial Action of Nitric Oxide-Releasing Chitosan Oligosaccharides against *Pseudomonas aeruginosa* under Aerobic and Anaerobic Conditions

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Chitosan oligosaccharides were modified with *N*-diazoniumdiolates to yield biocompatible nitric oxide (NO) donor scaffolds. The minimum bactericidal concentrations and MICs of the NO donors against *Pseudomonas aeruginosa* were compared under aerobic and anaerobic conditions. Differential antibacterial activities were primarily the result of NO scavenging by oxygen under aerobic environments and not changes in bacterial physiology. Bacterial killing was also tested against nonmucoid and mucoid biofilms and compared to that of tobramycin. Smaller NO payloads were required to eradicate *P. aeruginosa* biofilms under anaerobic versus aerobic conditions. Under oxygen-free environments, the NO treatment was 10-fold more effective at killing biofilms than tobramycin. These results demonstrate the potential utility of NO-releasing chitosan oligosaccharides under both aerobic and anaerobic environments.

Pseudomonas aeruginosa is an opportunistic human pathogen that frequently colonizes people with compromised immune systems, such as those with cystic fibrosis (CF) or severe burn wounds (1). The success of *P. aeruginosa* as a pathogen is related to its multitude of virulence factors, which increase adherence to the host cells, induce inflammation, and disrupt the host immune response (1). Furthermore, *P. aeruginosa* is intrinsically resistant to many antibiotics due to low membrane permeability and increased expression of β -lactamases and efflux pumps (2–4). In addition to this native resistance, *P. aeruginosa* readily adapts to antibiotic challenge by acquiring resistance genes (3, 5) and forming protective, cooperative communities known as biofilms (6, 7).

While all antibiotic resistance mechanisms are not fully understood, at least three main factors reduce antibiotic efficacy against bacterial biofilms compared to planktonic cells. First, *P. aeruginosa* in biofilms secretes a protective layer of exopolysaccharides that prevent the diffusion of antibiotics (7). In the context of cystic fibrosis, biofilm-bound *P. aeruginosa* exists predominantly as the mucoid phenotype, characterized by a secreted alginate matrix that provides a physical barrier against the host immune response and antibiotics (8). This exopolysaccharide matrix also prevents the diffusion of oxygen into biofilms, causing *P. aeruginosa* to switch from aerobic to anaerobic respiration (9). The reduced metabolic activity of *P. aeruginosa* undergoing anaerobic respiration protects the bacterium against traditional antibiotics that are most effective against rapidly dividing cells, including aminoglycosides and β -lactams (10, 11). Finally, biofilms produce persister cells (i.e., dormant bacteria that are highly resistant to chemical disinfectants and exhibit multidrug tolerance) much more frequently than planktonic bacterial cultures (3, 7).

The failure of conventional antibiotics to treat *P. aeruginosa* biofilms and infections necessitates the development of new antibacterial agents. Nitric oxide (NO), an endogenously produced free radical that can disperse (12, 13) and eradicate (14, 15) biofilms, holds particular promise as an alternative to current antibiotic treatments. Gaseous NO has been repeatedly used to eradicate *P. aeruginosa* infections in small-animal models with no apparent toxicity (16, 17). Under aerobic environments, NO reacts with

molecular oxygen, superoxide, and hydrogen peroxide to form highly reactive intermediates (peroxynitrite, nitrogen dioxide, and dinitrogen trioxide). These molecules exert nitrosative and oxidative stresses, such as DNA deamination, nitrosation of membrane and intracellular proteins, and membrane damage via lipid peroxidation, culminating in bacterial death (18–21). Some of these congener molecules, especially peroxynitrite, are more potent antimicrobials than NO alone (19). In anaerobic environments, NO toxicity is less understood. Ren et al. reported that the bacteriostatic mechanisms included modification of iron-sulfur proteins (22). As these proteins are linked to nearly every cellular process, including metabolism, respiration, RNA modification, and DNA repair and replication, their alteration greatly influences bacterial viability (23). The killing activities of NO are expected to be different under aerobic and anaerobic conditions due to the differences in bactericidal mechanisms, but this hypothesis has yet to be studied systematically. As *P. aeruginosa* grows rapidly and forms biofilms under both aerobic and anaerobic conditions, understanding the effects of oxygen on the antibacterial activity of NO is essential for developing NO-based therapeutics.

While the administration of exogenous NO holds promise as a therapeutic, treatment of infections or chronic wounds with gaseous NO is impractical, expensive, and potentially dangerous, as NO mediates other physiological processes (e.g., vasodilation and

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blood clotting) (21, 24–26). Macromolecular scaffolds capable of effectively storing and releasing NO have been developed to enable local delivery (27, 28). The most promising NO release vehicles to date include NO donor-modified *N*-diazoniumdiolate silica nanoparticles (29–31), dendrimers (32–36), and chitosan (15). While silica nanoparticles (14, 37–39) and dendrimers (32–34) are effective as antimicrobials, they do not easily break down and thus have limited potential as inhaled therapeutics. Chitosan-based oligosaccharides represent attractive scaffolds for NO delivery, as they are biodegradable and have low toxicity to mammalian cells (40, 41). We have previously reported that NO-releasing chitosan oligosaccharides are capable of NO storage/release and of eradicating *P. aeruginosa* biofilms under aerobic environments at concentrations nontoxic to mammalian cells (15). Here, we evaluate the antibacterial efficacy of NO-releasing chitosan oligosaccharides as a function of oxygen availability using nonmucoid, mucoid, and biofilm *P. aeruginosa* phenotypes.

MATERIALS AND METHODS

Materials. Medium-molecular-weight chitosan, 2-methylaziridine, and tobramycin were purchased from Sigma-Aldrich (St. Louis, MO). Methyltrimethoxysilane (MTMOS) was purchased from Fluka (Buchs, Switzerland). (Heptadecafluoro-1,1,2,2-tetrahydrodecyl)trimethoxysilane (17 FTMS) was purchased from Gelest (Morrisville, PA). Nitric oxide gas was purchased from Praxair (Sanford, NC). Standardized NO (26.85 ppm; balance N₂), argon (Ar), and nitrogen (N₂) gases were purchased from Airgas National Welders (Durham, NC). Sodium methoxide was purchased from Acros Organics (Geel, Belgium). Distilled water was purified using a Millipore Milli-Q UV Gradient A-10 system (Bedford, MA). All common laboratory salts and reagents were purchased from Fisher Scientific (Pittsburgh, PA). All materials were used without further purification unless otherwise specified.

Bacterial strains and media. The laboratory *P. aeruginosa* strain used in this study was strain K (PAK). The mucoid phenotype was the *muca22* isogenic mutant of the nonmucoid PAK strain. Both bacterial strains were a gift from Matthew Wolfgang, Department of Microbiology and Immunology, University of North Carolina (UNC) (Chapel Hill, NC). Clinical isolates were collected from patients at the UNC Hospital Clinical Microbiology Laboratory (Chapel Hill, NC). The clinical isolates were screened for tobramycin resistance using the Kirby-Bauer disk diffusion method according to standards published by the Clinical and Laboratory Standards Institute in document M100-S23 (42). All bacteria were grown in Luria Bertani (LB) broth (BD Biosciences, San Jose, CA) with the pH adjusted to 6.5 using 10 mM sodium phosphate. When indicated, potassium nitrate (15 mM) was added to the broth. Phosphate-buffered saline (PBS) was adjusted to pH 6.5 with 10 mM sodium phosphate. All bacterial media were adjusted to pH 6.5 to more accurately mimic the pH of CF mucus (22). Anaerobic media were kept in a Coy anaerobic chamber (Coy Laboratory Products, Ann Arbor, MI) with the lid loosened for 1 week prior to use.

Synthesis of COS. 2-Methylaziridine-modified chitosan oligosaccharides (COS) (specifically chitosan2-5k) were synthesized as previously described (15). Briefly, medium-molecular-weight chitosan (2.5 g) was oxidatively degraded to ~5 kDa in 15% hydrogen peroxide for 1 h at 85°C. The nondegraded chitosan was removed by filtration. The remaining chitosan oligosaccharides were precipitated from solution with acetone, collected via centrifugation, and dried *in vacuo*. The ~5-kDa chitosan oligosaccharides (500 mg) were then dissolved in water (10 ml). Concentrated hydrochloric acid (27.6 μl), water (250 μl), and 2-methylaziridine (356 μl) were then added to this solution. The reaction mixture was stirred for 5 days at 25°C, followed by 24 h at 75°C. The modified chitosan oligosaccharides were again precipitated with acetone, collected via centrifugation, and dried *in vacuo*. The ¹H nuclear magnetic resonance (NMR) data for COS were (400 MHz, D₂O,

δ): 0.8–1.1 [NH₂CH(CH₃)CH₂NH], 1.9 (C-7: CHNHCOCH₃), 2.3–2.9 [NH₂CH(CH₃)CH₂NHCH, C-2: NH₂CH(CH₃)CH₂NHCH], 3.3–4.0 [C-3, C-4, C-5, C-6: OHCH, OCHCH(OH)CH(NH₂)CH, OHCH₂CH, OHCH₂CH], and 4.4 [C-1: OCH(CHNH₂)O].

Synthesis of COS-NO. In order to impart NO storage and release, *N*-diazoniumdiolates were formed on the secondary amines of COS (15). Briefly, COS (15 mg) was dissolved in a solution of water (300 μl), methanol (700 μl), and 5.4 M sodium methoxide (25 μl) in a 1-dram vial equipped with a stir bar. The open vial was placed in a 160-ml Parr general purpose stainless steel pressure vessel and rigorously stirred. Oxygen was removed from the reaction vessel by purging with argon (10 s; 8 × 10⁵ Pa) 3 times, followed by 3 additional long argon purges (10 min; 8 × 10⁵ Pa). The vessel was then filled with potassium hydroxide-purified NO gas (10 × 10⁵ Pa) for 72 h at room temperature. Afterward, the argon-purging procedure was repeated to remove unreacted NO. The *N*-diazoniumdiolate-modified chitosan oligosaccharides (COS-NO) were precipitated in acetone, collected via centrifugation, dried *in vacuo*, and stored at –20°C as a yellow powder.

Chemiluminescence detection of NO release. A Sievers (Boulder, CO) 280i chemiluminescence nitric oxide analyzer was used for chemiluminescence detection of NO from COS-NO (1.0 mg) in 30 ml of deoxygenated PBS (pH 6.5) at 37°C. The released NO was carried by N₂ gas to the reaction vessel/detector at a flow rate of 80 ml/min. Additional N₂ flow was supplied to the sample flask at 200 ml/min to match the collection rate of the instrument. The analysis was terminated when NO concentrations fell below 10 ppb NO/mg COS-NO. Prior to analysis, the instrument was calibrated with air passed through an NO zero filter (0 ppm NO) and 26.8 ppm of NO standard gas (balance N₂).

Electrochemical detection of NO release. NO-selective electrochemical sensors were fabricated in house as previously reported (43). Briefly, polished polycrystalline Pt disk electrodes (2 mm) sealed in Kel-F (CH Instruments, Austin, TX) were coated with an NO-selective membrane prepared by mixing MTMOS (60 μl), ethanol (300 μl), 17 FTMS (15 μl), water (80 μl), and 0.5 M hydrochloric acid (5 μl) for 1 h at 25°C. The resulting solution was spreadcast over the Pt electrode and dried overnight at room temperature. Amperometric NO measurements followed, using a three-electrode setup with the NO-selective membrane-modified Pt electrode as the working electrode, a Pt-coiled counter electrode, and an Ag/AgCl reference electrode. The applied potential for NO oxidation was +700 mV versus Ag/AgCl. Immediately prior to use, the NO sensors were calibrated by adding a known amount of PBS saturated with NO gas (1.9 mM) to deoxygenated PBS (pH 6.5). Saturated NO solutions were made on the day of use by degassing PBS (pH 6.5) for 30 min with Ar, followed by 20-min purging with NO gas. The sensors were immersed in 10.0 ml of PBS or LB broth (stirred; 37°C) and polarized at +700 mV versus Ag/AgCl until a stable baseline was achieved prior to the addition of COS-NO. The NO oxidation current was measured every 0.1 s and ceased when the current returned to its background value. Measurement of NO release in anaerobic media was carried out in a Coy anaerobic chamber. Total NO for 1.0-mg COS-NO/ml solutions are reported as the average and standard deviation for 4 or more separate measurements.

Planktonic bactericidal assays. Bacteria were grown as overnight cultures, diluted 1:100 in fresh LB broth (with or without nitrate supplementation), and grown to mid-log phase (2 × 10⁸ CFU/ml). These cultures were centrifuged, resuspended in PBS, and diluted to 2 × 10⁶ CFU/ml in PBS. Each suspension was then added to vials containing 2-fold serial dilutions of COS-NO or COS controls and incubated at 37°C for 4 h with gentle shaking. Following treatment, the bacterial solutions were serially diluted, spiral plated on LB agar, and incubated for 24 h at 37°C. Colonies were enumerated using a Flash and Go colony counter (IUL, Farmingdale, NY). The minimum bactericidal concentration after a 4-h exposure (MBC_{4h}) was defined as the minimum concentration required to achieve a 3-log-unit reduction in viable bacteria (from 10⁶ to 10³ CFU/ml). The plating-counting method employed has a limit of detection of 2.5 × 10³ CFU/ml (44). The corresponding NO dose was calculated by multiplying

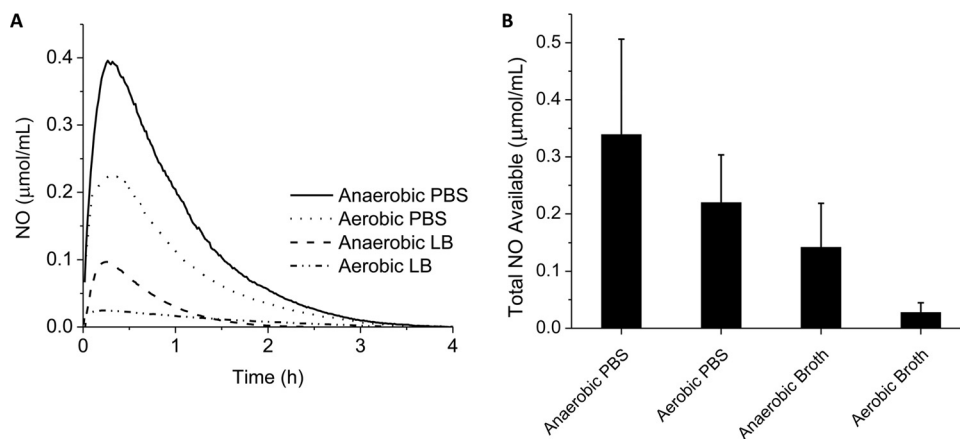


FIG 1 Electrochemical measurements of available NO in media. (A) Representative NO release profiles for 1.0 mg/ml of COS-NO in anaerobic PBS, aerobic PBS, anaerobic LB broth, and aerobic LB broth. (B) The values from panel A were integrated to find the total concentrations of available NO in a 1.0-mg/ml solution of COS-NO in biological media after 4 h. The error bars indicate standard deviations.

the MBC_{4h} of COS-NO (in milligrams per milliliter) with the available NO in aerobic and anaerobic PBS (in micromoles NO per milligram COS-NO).

Planktonic inhibition assays. Bacteria were grown as overnight cultures, diluted 1:100 in fresh LB broth, grown to mid-log phase (2×10^8 CFU/ml), and diluted to 2×10^6 CFU/ml in LB broth. The bacterial cultures were then added to vials containing 2-fold serial dilutions of COS-NO or COS controls and incubated at 37°C for 18 h with gentle shaking. The MIC was determined to be the minimum concentration that prevented visible growth, defined as an optical density at 600 nm of <0.1 . All untreated (control) cultures became visibly turbid during the 18-h growth period. The corresponding NO dose was calculated by multiplying the MIC of COS-NO (in milligrams per milliliter) with the available NO in aerobic and anaerobic broth (in micromoles NO per milligram COS-NO). Nitrate-supplemented LB was used for all stages of bacterial growth and exposure. Anaerobic experiments were performed in a Coy anaerobic chamber.

Biofilm eradication assays. Bacteria were grown as overnight cultures, diluted 1:100 in fresh LB broth, and grown to mid-log phase (2×10^8 CFU/ml). The bacterial cultures were then diluted to 10^6 CFU/ml in diluted (25%) LB broth supplemented with 15 mM KNO_3 (pH 6.5) and grown for 72 h at 37°C with gentle shaking. The viscous microcolony biofilms formed were easily separated from the growth media via pipetting. The biofilms were harvested by placing a pipette tip near the center of the biofilm and applying suction. The biofilms were then washed by injection into PBS and extracted using the same pipetting procedure to remove planktonic or loosely associated bacteria. The freshly washed biofilms (250 μ l) were combined with 750 μ l of PBS (pH 6.5); added to vials containing COS, COS-NO, or tobramycin; and incubated with gentle shaking for 18 h at 37°C. After treatment, the biofilms were washed via pipetting in PBS to remove excess antibacterial agent, transferred to 750 μ l of PBS (pH 6.5), and gently sonicated to disrupt the biofilm matrix. The dispersed biofilms were vortexed, serially diluted, plated, and enumerated on LB agar. The minimum biofilm eradication concentration at 18 h ($MBEC_{18h}$) was defined as the concentration that caused a 5-log-unit reduction in viable bacteria (i.e., 10^8 to 10^3 CFU/ml) after the 18-h treatment. The corresponding NO dose was calculated by multiplying the $MBEC_{18h}$ of COS-NO (in milligrams per milliliter) with the available NO in aerobic and anaerobic PBS (in micromoles NO per milligram COS-NO).

Statistical analysis. All data are expressed as the mean \pm 1 standard deviation and were analyzed for significance ($P < 0.05$) with a two-tailed Student *t* test.

RESULTS

Nitric oxide release from COS-NO in media. Nitric oxide release from COS-NO was measured via chemiluminescence in deoxygenated PBS (pH 6.5) at 37°C to yield total NO release payloads of 0.86 ± 0.05 μ mol NO/mg, with an overall release duration of 10.2 ± 2.7 h (see Fig. S1 in the supplemental material). While NO release from macromolecular scaffolds is generally measured in deoxygenated medium, measuring the amount of bioavailable (i.e., nonscavenged) NO is critical for elucidating the biocidal dose-response relationship of NO under the intended conditions, as NO is rapidly scavenged by oxygen and proteins in biological media (45). Unfortunately, foaming associated with nutrient-rich medium makes chemiluminescence detection difficult and irreproducible (43). Thus, we turned to amperometric NO detection to carry out NO measurements in broth (LB).

In the absence of NO scavenging (i.e., in deoxygenated PBS), amperometric measurements revealed a total NO payload of 0.34 ± 0.17 μ mol/ml from the 1.0-mg/ml solution of COS-NO over 4 h (Fig. 1). As might be expected, both the total NO payload and the release duration of COS-NO measured via amperometry were reduced compared to chemiluminescence detection. These decreases are common for electrochemical sensors that are based on the diffusion of NO to the working electrode and the inherent loss of NO to the ambient atmosphere (43).

Under aerobic conditions, oxygen scavenging reduced the amount of free NO available in PBS by approximately 35% (0.22 ± 0.08 μ mol NO/ml). Nutrient broth (LB) further diminished the available NO payload via scavenging of the NO by proteins in the broth. The amount of NO available in anaerobic LB broth was reduced to 0.14 ± 0.08 μ mol NO/ml, a 66% reduction relative to anaerobic PBS. Further reductions (0.027 ± 0.017 μ mol/ml) were observed in aerobic LB broth due to reaction of NO with oxygen.

Bactericidal action of the COS scaffold. To confirm that NO and not the scaffold was responsible for the observed bacterial killing, all bacterial assays were performed using NO-releasing and control (i.e., non-NO-releasing) chitosan oligosaccharides. In MBC_{4h} assays of planktonic cells, COS did not influence bacterial viability at $1 \times$ or $10 \times$ the MBC_{4h} of COS-NO, indicating that the

TABLE 1 Effects of oxygen on nongrowing planktonic cultures^a

Strain	Growth medium	Aerobic exposure		Anaerobic exposure	
		MBC _{4h} (μg COS-NO/ml)	NO dose ^b (μmol NO/ml)	MBC _{4h} (μg COS-NO/ml)	NO dose ^b (μmol NO/ml)
Nonmuroid	Aerobic	100	0.022 ± 0.008	100	0.034 ± 0.017
	Anaerobic	100	0.022 ± 0.008	100	0.034 ± 0.017
Muroid	Aerobic	100	0.022 ± 0.008	100	0.034 ± 0.017
	Anaerobic	200	0.044 ± 0.016	200	0.068 ± 0.033

^a *P. aeruginosa* cultures were grown in LB broth (plus 15 mM KNO₃) under aerobic or anaerobic conditions and then exposed to COS-NO in PBS (pH 6.5) for 4 h under aerobic or anaerobic conditions.

^b Determined via amperometry. The values are presented as means ± standard deviations for 3 or more pooled experiments.

chitosan oligosaccharide alone was not bactericidal (see Fig. S2 in the supplemental material). Similarly, bacterial viability was not reduced upon treatment of the biofilms with 4.0 mg COS/ml (1× the MBEC_{18h}) for 18 h under both aerobic and anaerobic conditions (see Fig. S3 in the supplemental material). Based on these data, the bactericidal activity of COS-NO was attributed solely to the effects of NO and not to toxicity of the COS scaffold.

Effect of oxygen on bactericidal action of NO against planktonic *P. aeruginosa*. The biocidal action of NO was evaluated with respect to the oxygen concentration in the treatment medium by exposing planktonic cultures to COS-NO in both aerobic and anaerobic PBS (pH 6.5) (Table 1). When grown under aerobic conditions, the same concentration of COS-NO (100 μg/ml) was required to eradicate both the muroid and nonmuroid strains. Furthermore, oxygen in the exposure medium did not alter the bactericidal concentration of COS-NO. However, the NO dose delivered was slightly greater under anaerobic exposure conditions due to the decreased reactions between NO and oxygen. For example, the bactericidal NO dose was 0.022 ± 0.008 μmol NO/ml under aerobic conditions versus 0.034 ± 0.017 μmol NO/ml in deoxygenated (anaerobic) medium. This difference in the NO payload was not statistically significant.

Bacterial cultures were also grown aerobically and anaerobically to determine if oxygen in the growth medium affected *P. aeruginosa* susceptibility to NO. As anaerobic growth requires nitrate, all assays were carried out using nitrate-supplemented LB medium to enable direct comparison. The absence of oxygen in the growth medium had no effect on the susceptibility of nonmuroid *P. aeruginosa* to NO. However, strict anaerobic growth of muroid *P. aeruginosa* increased the tolerance of the strain for NO by 2-fold (MBC_{4h} = 0.044 ± 0.016 and 0.022 ± 0.008 μmol NO/ml for anaerobic and aerobic growth conditions, respectively) (Table 1). The increased tolerance for NO was observed under both aerobic and anaerobic exposure conditions.

Inhibition of planktonic *P. aeruginosa* growth by COS-NO. MIC assays were performed to evaluate the efficacy of COS-NO

during bacterial growth under aerobic and anaerobic environments (Table 2). Under aerobic conditions, the nonmuroid phenotype was more tolerant of COS-NO than the muroid strain, with inhibitory doses of 800 μg COS-NO/ml (0.022 ± 0.014 μmol NO/ml) versus 400 μg COS-NO/ml (0.011 ± 0.007 μmol NO/ml), respectively. Anaerobic conditions decreased the MIC to 100 μg COS-NO/ml (0.014 μmol NO/ml) for both phenotypes. While the COS-NO concentration required to inhibit growth was reduced in anaerobic environments, the NO dose delivered was not significantly lower, indicating that NO lost to reaction with oxygen accounts for the increased MICs against COS-NO under aerobic conditions.

Inhibition of growth by COS-NO for clinical isolates, including tobramycin-resistant strains. To ensure that the increased inhibition of bacterial growth by COS-NO under anaerobic conditions was not solely a function of the laboratory *P. aeruginosa* strain used, 10 clinical isolates of *P. aeruginosa* were tested, including muroid, nonmuroid, and tobramycin-resistant isolates. The concentrations of COS-NO required to inhibit growth under anaerobic conditions were less than or equal to those under aerobic conditions for all 10 isolates (Fig. 2A). There was no statistical difference in the NO dose required to inhibit growth for most of the strains tested (Fig. 2B). However, two isolates (Fig. 2B, asterisks) showed a statistically significant increase in the NO dose required to inhibit growth under anaerobic conditions relative to aerobic conditions. Overall, COS-NO was more effective at inhibiting growth under anaerobic environments, but the efficacy of NO was unchanged for most of the isolates tested.

As with the laboratory strain, differences were seen between the muroid and nonmuroid phenotypes (Fig. 2). Muroid strains were more susceptible to COS-NO, as indicated by the low MIC range (25 to 200 μg COS-NO/ml) relative to nonmuroid strains (MIC range, 100 to 400 μg COS-NO/ml). No apparent differences were observed between tobramycin-susceptible and tobramycin-resistant strains of the same phenotype; however, more isolates would be needed to confirm the statistical significance of these trends.

TABLE 2 Influence of oxygen on the inhibitory efficacy of COS-NO^a

Growth medium	Nonmuroid strain		Muroid strain	
	MIC (μg COS-NO/ml)	NO dose ^b (μmol NO/ml)	MIC (μg COS-NO/ml)	NO dose ^b (μmol NO/ml)
Aerobic	800	0.022 ± 0.014	400	0.011 ± 0.007
Anaerobic	100	0.014 ± 0.008	100	0.014 ± 0.008

^a Bacterial cultures in mid-log-phase growth were diluted to 2 × 10⁶ CFU/ml in LB broth (plus 15 mM KNO₃) with COS-NO and grown for 18 h under aerobic or anaerobic conditions. The MIC was determined as the concentration of COS-NO that visibly inhibited growth.

^b Determined via amperometry. The values are presented as means ± standard deviations for 3 or more pooled experiments.

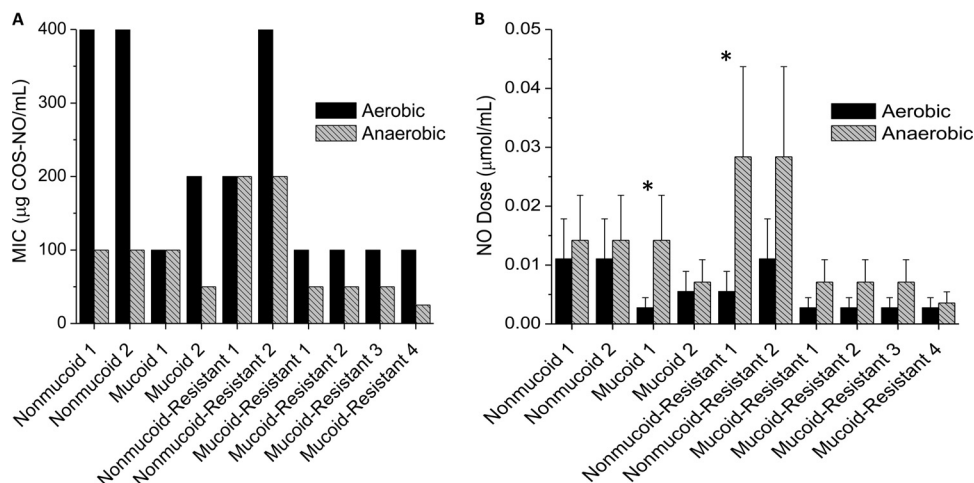


FIG 2 MICs against clinical isolates of *P. aeruginosa*. (A) Minimum concentrations of COS-NO resulting in no bacterial growth for nonmucoid, mucoid, and tobramycin-resistant isolates. (B) Corresponding NO dose for each isolate under aerobic and anaerobic environments. All bacteria were grown and exposed in LB medium supplemented with nitrate (pH 6.5) for 18 h. Statistically significant differences ($P < 0.05$) between aerobic and anaerobic NO payloads are indicated by the asterisks. The error bars indicate standard deviations.

Biofilm eradication by COS-NO and tobramycin. As bacterial biofilms exist in both aerobic and anaerobic environments (46), it was important to determine how oxygen concentrations affected the antibiofilm activity of COS-NO. Under aerobic conditions, highly viscous microcolony biofilms were formed ($\sim 250 \mu\text{l}$ in volume) with bacterial viability of $4.0 \pm 0.6 \times 10^8$ and $2.5 \pm 0.5 \times 10^8$ CFU/ml for nonmucoid and mucoid phenotypes, respectively. Of note, nitrate supplementation was required to prevent phenotypic switching from the mucoid to the nonmucoid phenotype (47). Under anaerobic growth conditions, we were unable to cause the bacteria to form robust biofilms even after 7 days of growth.

Bacterial biofilms were exposed to COS-NO for 18 h in PBS (pH 6.5) under aerobic or anaerobic conditions. The MBEC_{18 h} values for the two phenotypes were 4,000 μg COS-NO/ml and 1,000 μg COS-NO/ml (0.88 ± 0.33 and $0.34 \pm 0.17 \mu\text{mol}$ NO/ml) under aerobic and anaerobic conditions, respectively (Table 3). These results indicate that NO released from a scaffold is equally effective at eliminating biofilms derived from nonmucoid and mucoid strains. Moreover, NO is significantly more effective at eliminating biofilms in the absence of oxygen.

The MBEC_{18 h} of tobramycin against biofilms was determined under the same conditions (i.e., 18-h exposure in PBS) to allow comparison of NO to current antibiotic therapies. Under aerobic environments, the nonmucoid strain was eradicated at lower concentrations of tobramycin than the mucoid strain (200 and 800 $\mu\text{g}/\text{ml}$, respectively) (Table 4). Both strains required greater to-

bramycin levels (1,600 $\mu\text{g}/\text{ml}$) to eradicate bacterial biofilms under anaerobic conditions.

DISCUSSION

We have previously reported on the antibacterial activity of NO against planktonic and biofilm-based *P. aeruginosa* (14, 15, 39). However, little is understood regarding how oxygen and the bacterial phenotype impact NO's efficacy. Such knowledge is critical in the development of NO-based therapeutics. Water-soluble NO-releasing chitosan oligosaccharides were used as the NO release scaffold in the studies described here due their biocompatibility and ability to be degraded *in vivo* (40, 41, 48). Although chitosan is a known bactericidal agent, the reduced molecular mass (to ensure water solubility) and 2-methylaziridine modification (for NO donor addition) resulted in a material with no bactericidal activity (see Fig. S2 and S3 in the supplemental material).

It is well known that NO reacts with oxygen and superoxide to form highly reactive intermediates that facilitate bacterial killing through oxidative and nitrosative stresses (19, 49). As oxygen plays an integral role in the antibacterial action of NO, anaerobic environments may reduce the biocidal efficacy of NO (20, 50). However, NO also reacts with oxygen to form nitrate and nitrite. These seemingly paradoxical roles of oxygen in NO-mediated killing are not fully understood. Therefore, we carried out electrochemical measurements of NO under aerobic and anaerobic conditions to quantify the amounts of bioavailable NO. Under

TABLE 3 Bactericidal efficacy of COS-NO against *P. aeruginosa* biofilms^a

Strain	Aerobic exposure		Anaerobic exposure	
	MBEC (μg COS-NO/ml)	NO dose ^b (μmol NO/ml)	MBEC (μg COS-NO/ml)	NO dose ^b (μmol NO/ml)
Nonmucoid	4,000	0.88 ± 0.33	1,000	0.34 ± 0.17
Mucoid	4,000	0.88 ± 0.33	1,000	0.34 ± 0.17

^a Biofilms were exposed to COS-NO in PBS (pH 6.5) for 18 h under aerobic or anaerobic conditions. The MBEC_{18 h} is reported as the concentration of COS-NO required for 5-log-unit reduction in biofilm viability.

^b Determined via amperometry. The values are presented as means \pm standard deviations for 3 or more pooled experiments.

TABLE 4 Bactericidal efficacy of tobramycin against *P. aeruginosa* biofilms^a

Strain	MBEC [$\mu\text{g/ml}$ ($\mu\text{mol/ml}$)]	
	Aerobic exposure	Anaerobic exposure
Nonmucoid	200 (0.43)	1,600 (3.42)
Mucoid	800 (1.71)	1,600 (3.42)

^a Biofilms were exposed to tobramycin in PBS (pH 6.5) for 18 h under aerobic or anaerobic conditions. The MBEC_{18 h} is reported as the concentration of COS-NO required for 5-log-unit reduction in biofilm viability.

aerobic conditions, the measured NO decreased by 35% compared to anaerobic conditions (Fig. 1). To elucidate the effects of oxygen availability in treatment media, bacteria were first grown aerobically and then exposed to COS-NO in aerobic or anaerobic PBS. Identical concentrations of COS-NO were required to kill *P. aeruginosa* regardless of the treatment conditions. Due to the reaction of NO with oxygen, the bioavailable concentration of NO (i.e., the NO dose) was slightly, but not significantly, higher under anaerobic conditions (Table 1). As such, oxygen availability in the treatment medium has no statistically significant effect on the biocidal activity of NO released from COS-NO.

While the oxygen concentration in the exposure medium did not alter the bactericidal efficacy of NO, the presence of oxygen during bacterial growth did influence *P. aeruginosa* susceptibility to NO. Anaerobic growth conditions reduce the efficacy of current antibiotics by altering certain properties of the bacteria, such as alginate production (46) and metabolic rates (10). To separate these factors, MBC_{4 h} assays were performed under nonnutritive conditions to minimize the effects of bacterial metabolism on the bactericidal activity of NO. When bacteria were grown under anaerobic conditions, the efficacy of NO released from the chitosan oligosaccharide scaffold was decreased against the mucoid, but not the nonmucoid, phenotype, indicating that growing mucoid bacteria without oxygen significantly alters their defense against NO (Table 1). Worlitzsch et al. previously reported that *P. aeruginosa* produces a protective alginate exopolysaccharide that is 50% thicker when grown anaerobically (46). As alginate restricts the diffusion of oxygen, the increased thickness of this protective layer could potentially prevent NO diffusion into the bacteria, therefore requiring a larger NO dose for killing.

To study the role of anaerobic growth on the efficacy of NO and COS-NO, we evaluated the inhibition of *P. aeruginosa* growth by COS-NO in nutrient-rich medium under both aerobic and anaerobic conditions. In contrast to the static conditions of MBC_{4 h} assays, bacteria are actively growing during inhibition assays. Comparison of MIC values under oxygen and oxygen-free environments showed that the efficacy of COS-NO was enhanced under anaerobic conditions while there was no statistical difference in the corresponding NO dose. This behavior was observed in the laboratory strains (Table 2) and most of the clinical isolates tested (Fig. 2). As the efficacy of NO is not reduced under anaerobic environments, NO-based treatments represent a potential alternative to current antibiotic treatments, including aminoglycosides and β -lactams, which are less effective under anaerobic conditions because their mechanism of action requires actively dividing cells (51, 52).

It is important to characterize the antibiofilm activities of antibacterial agents, as *P. aeruginosa* exists as biofilms on medical

implants (e.g., catheters) (53, 54), on burn wounds (55, 56), and in the airways of patients with cystic fibrosis (57, 58). As shown in Table 4, mucoid biofilms are significantly more resilient against tobramycin than nonmucoid biofilms. Hentzer et al. attributed decreased antibiotic efficacy against mucoid strains to the overproduction of alginate (59). As has been previously reported (60, 61), low-oxygen conditions further decrease the effectiveness of tobramycin (the MBEC_{18 h} is increased to 1,600 $\mu\text{g/ml}$ under anaerobic conditions). While tobramycin is a highly effective antipseudomonal agent, these factors compromise its ability to kill *P. aeruginosa* biofilms in oxygen-free environments. In contrast, NO released from the chitosan scaffold was equally effective at eradicating mucoid and nonmucoid biofilms. Similarly, the antibiofilm activity was not reduced under anaerobic environments (Table 3). Furthermore, the NO dose required for biofilm eradication under anaerobic conditions is $0.34 \pm 0.17 \mu\text{mol NO/ml}$, 10-fold lower than that of tobramycin (3.42 $\mu\text{mol/ml}$) (Tables 3 and 4).

In conclusion, these studies examined the susceptibilities of nonmucoid, mucoid, and biofilm *P. aeruginosa* phenotypes to NO-releasing chitosan oligosaccharides as a function of oxygen availability. The antibacterial activity of NO-releasing chitosan oligosaccharides was enhanced in oxygen-free environments, despite a concomitant decrease in the number of possible mechanisms available to kill bacteria (i.e., fewer toxic by-products from the reactions of NO and oxygen). Furthermore, the antibiofilm action of NO was more effective than that of tobramycin and was not influenced by the bacterial phenotype. When combined with NO's significant biocidal action against *P. aeruginosa*, these results suggest that NO-releasing chitosan oligosaccharides may represent a potential alternative to traditional antibiotics, particularly when treating biofilms or in low-oxygen environments. We are currently seeking to enhance the NO payloads and evaluate the effects of NO release kinetics on the antibacterial efficacy of chitosan oligosaccharides.

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Mark H. Schoenfisch declares a competing financial interest: he is a cofounder and member of the board of directors and maintains a financial interest in Novan Therapeutics, Inc. Novan Therapeutics, Inc. provided partial funding for this research and is commercializing macromolecular nitric oxide storage and release vehicles for dermatological applications.

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