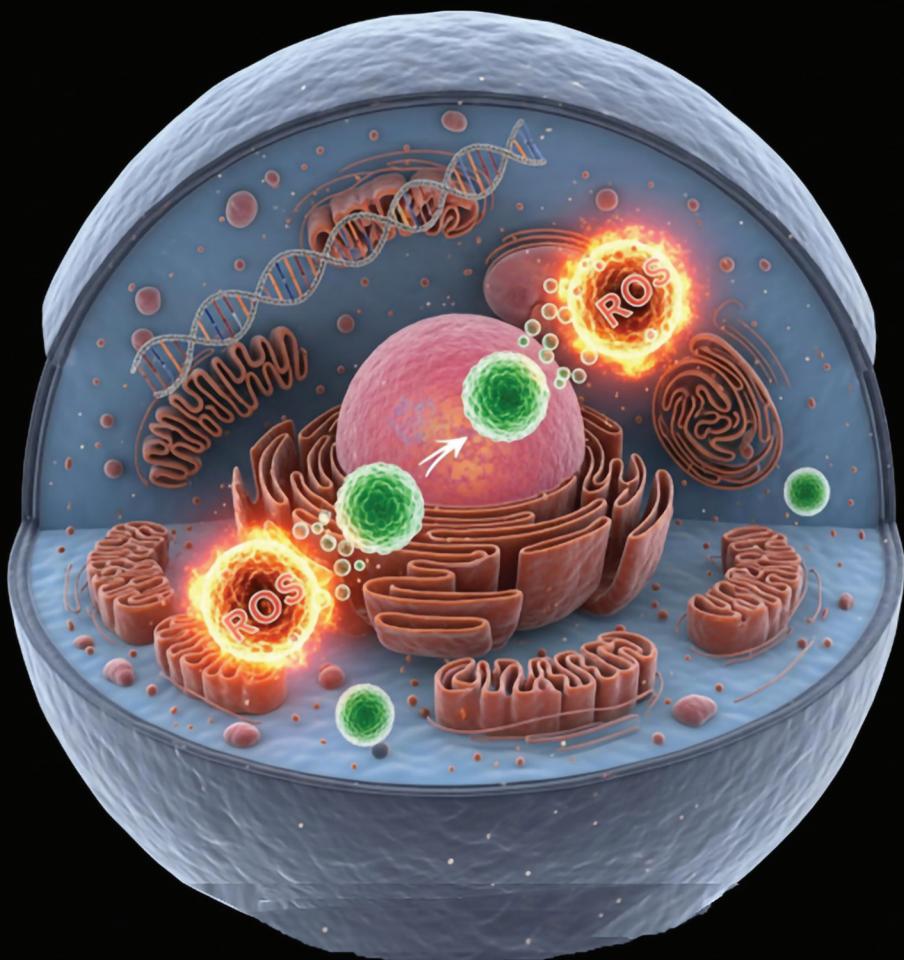


Nanoscale

rsc.li/nanoscale



ISSN 2040-3372


 Cite this: *Nanoscale*, 2025, **17**, 18077

 Received 29th January 2025,
 Accepted 24th April 2025
 DOI: 10.1039/d5nr00430f

rsc.li/nanoscale

CeO₂- and Mn₃O₄-based nanozymes exhibit scavenging of singlet oxygen species and hydroxyl radicals†

 Krishnendu M. R., ^{a,b} Diyya Mehta^{a,b} and Sanjay Singh ^{*a,b}

Singlet oxygen and hydroxyl radicals are highly reactive species that contribute significantly to oxidative stress-related pathologies. Herein, we report the effective scavenging of $^1\text{O}_2$ and $^{\cdot}\text{OH}$ by CeO₂- and Mn₃O₄-based nanozymes and compare them with the well-known scavengers of these radicals. The IC₅₀ values of scavenging of $^{\cdot}\text{OH}$ by these nanozymes were compared with NAC, which were in the order of Mn₃O₄ [2.3 μM] < CeO₂ [5.4 μM] < NAC [28.4 μM]. Similarly, the IC₅₀ values for $^1\text{O}_2$ scavenging were in the order of Mn₃O₄ [4.75 μM] < sodium azide [60.5 μM] < CeO₂ [857.27 μM]. The cell viability assays, ROS generation studies and cell cycle analysis revealed that these nanozymes (1 $\mu\text{g mL}^{-1}$) are biocompatible with mammalian cells.

Reactive oxygen species (ROS) generated through the chemical, photochemical, or biochemical reduction of oxygen are redox active intermediates that trigger a series of oxidative reactions.¹ Among various ROS, superoxide anions ($\text{O}_2^{\cdot-}$), hydroxyl radicals ($^{\cdot}\text{OH}$), singlet oxygen ($^1\text{O}_2$), and hydrogen peroxide (H_2O_2) are well known to orchestrate various biochemical reactions in mammalian cells/tissues. Overproduction of these ROS causes oxidation of various biological macromolecules, leading to critical ailments such as cancer, cardiovascular and neurodegenerative diseases, *etc.* Various non-invasive cancer treatment strategies such as photothermal, photodynamic, and sonodynamic therapies involve the generation of excessive ROS for localized damage of cancer tissues.^{2–4} Nanozymes have also been utilized as oxygen-independent radiodynamic therapeutics and also as pro-drugs for the treatment of various diseases.^{5,6} Several synthetic scavengers, such as corticosteroids and nonsteroidal anti-inflammatory drugs, have been used to alleviate the toxic effects of ROS; however, they cause

gastrointestinal and cardiovascular complications and renal failure.⁷ Recently, antioxidant nanozymes such as cerium oxide (CeO₂ NPs) and manganese oxide (Mn₃O₄ NPs) and derivatives of fullerenes have been discovered to scavenge the extremely reactive and harmful $\text{O}_2^{\cdot-}$ and H_2O_2 .^{8,9} Although a lot of effort has been made to develop scavengers of $\text{O}_2^{\cdot-}$ and H_2O_2 , limited attention is given to $^{\cdot}\text{OH}$ and $^1\text{O}_2$ scavenging. These radicals are particularly significant due to their extreme reactivity and potential for causing severe cellular damage, leading to various diseases.

The well-explored methods of $^{\cdot}\text{OH}$ generation are the Fenton and Haber–Weiss reactions, which contribute to various pathologies, including liver damage in obstructive jaundice, ascites syndrome, familial amyotrophic lateral sclerosis (FALS), and ischemia-induced intestinal vascular permeability.^{10–13} $^1\text{O}_2$, an electronically excited form of oxygen, plays a key role in oxidative stress, particularly in conditions like acute endotoxemia and type 2 diabetes, leading to impaired glucose tolerance and insulin resistance.^{14,15} Given the strong oxidizing nature of $^{\cdot}\text{OH}$ and $^1\text{O}_2$, it is highly desirable to develop materials that can effectively scavenge these radicals from biological systems to prevent oxidative damage. CeO₂ and Mn₃O₄ NPs have been extensively studied for their antioxidant properties to scavenge ROS, such as $\text{O}_2^{\cdot-}$ and H_2O_2 . However, there are fewer reports on scavenging $^{\cdot}\text{OH}$ and $^1\text{O}_2$ by nanozymes. Reported methods for nanozyme-mediated ROS scavenging often rely on complex processes of radical generation or detection, exhibit limited efficiency, and are effective under acidic conditions.^{16–18} Additionally, scavengers of $^1\text{O}_2$ such as sodium azide (NaN₃) raise toxicity concerns due to the inhibition of metalloenzymes,¹⁹ while $^{\cdot}\text{OH}$ scavenging polyamines require relatively high concentrations to achieve effective neutralization.²⁰ These limitations highlight the need for developing biocompatible and efficient nanozymes capable of scavenging reactive species under physiologically relevant conditions. Herein, we report the synthesis of dextran-coated CeO₂ and Mn₃O₄ NPs/nanozymes that are biocompatible and efficient at scavenging $^{\cdot}\text{OH}$ and $^1\text{O}_2$. We report novel methods to generate these radicals and employ multiple independent

^aNanobiology and Nanozymology Research Laboratory, National Institute of Animal Biotechnology (NIAB), Opposite Journalist Colony, Near Gowlidoddy, Extended Q-City Road, Gachibowli, Hyderabad, Telangana 500032, India.

E-mail: sanjay@niab.org.in

^bRegional Centre for Biotechnology (RCB), Faridabad 121001, Haryana, India

† Electronic supplementary information (ESI) available. See DOI: <https://doi.org/10.1039/d5nr00430f>

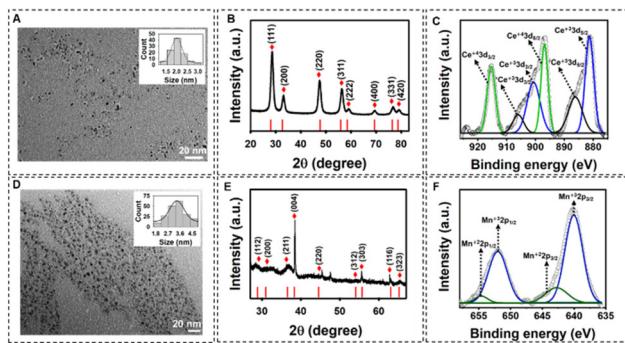


Fig. 1 Characterization of Dex-CeO₂ NPs and Dex-Mn₃O₄ NPs: (A) TEM image (inset shows particle size distribution), (B) XRD pattern and (C) XPS spectra of Dex-CeO₂ NPs. (D) TEM image (inset shows particle size distribution), (E) XRD pattern and (F) XPS spectra of Dex-Mn₃O₄ NPs.

experiments to confirm the scavenging capabilities of these nanozymes and propose the possible reaction mechanisms.

Dextran-coated cerium oxide nanoparticles (Dex-CeO₂ NPs) and manganese oxide nanoparticles (Dex-Mn₃O₄ NPs) were characterized by several advanced techniques. FTIR spectra revealed characteristic transmissions at 450 cm⁻¹ and 1380 cm⁻¹ for Dex-CeO₂ NPs, corresponding to Ce–O stretching and vibrational modes (Fig. S1A†),²¹ while Dex-Mn₃O₄ NPs showed signals at 519 and 613 cm⁻¹, corresponding to Mn–O and Mn–O–Mn bond vibrations (Fig. S2A†).²² The UV-Vis spectra of both nanoparticles showed a peak at 290 nm, confirming the dextran coating (Fig. S1B and S2B†). TEM analysis revealed that Dex-CeO₂ NPs have an average size of ~2 nm (Fig. 1A), while Dex-Mn₃O₄ NPs were ~3 nm in diameter (Fig. 1D). XRD analysis of Dex-CeO₂ NPs showed diffraction peaks matching the CeO₂ fluorite structure (JCPDS file no. 34-0394) (Fig. 1B). XRD analysis confirmed the tetragonal phase of Mn₃O₄ (JCPDS file no. 24-0734) (Fig. 1E). XPS analysis of Dex-CeO₂ NPs revealed the presence of both Ce³⁺ and Ce⁴⁺ states. The Ce 3d core-level spectrum for Ce⁴⁺ was deconvoluted into a single spin-orbit pair (915.25 eV and 896.85 eV), while Ce³⁺ exhibited two spin-orbit pairs (900.75 eV/881.4 eV and 906.00 eV/886.18 eV) representing 3d_{5/2} and 3d_{3/2}, respectively (Fig. 1C).²³ The integrated area under the two spin-orbit pairs of Ce³⁺ and the single spin-orbit pair of Ce⁴⁺ was used to calculate the Ce³⁺/Ce⁴⁺ ratio and *vice versa*^{24,25} (Table S1†). The following equations were used for the calculation of [Ce³⁺]:

$$[\text{Ce}^{3+}] = \frac{[\text{ACe}^{3+}3\text{d}_{5/2} + \text{ACe}^{3+}3\text{d}_{3/2} + \text{A}'\text{Ce}^{3+}3\text{d}_{5/2} + \text{A}'\text{Ce}^{3+}3\text{d}_{3/2}]}{[\text{ACe}^{3+}3\text{d}_{5/2} + \text{ACe}^{3+}3\text{d}_{3/2} + \text{A}'\text{Ce}^{3+}3\text{d}_{5/2} + \text{A}'\text{Ce}^{3+}3\text{d}_{3/2} + \text{ACe}^{4+}3\text{d}_{5/2} + \text{ACe}^{4+}3\text{d}_{3/2}]}$$

and [Ce⁴⁺]:

$$[\text{Ce}^{4+}] = \frac{[\text{ACe}^{4+}3\text{d}_{5/2} + \text{ACe}^{4+}3\text{d}_{3/2}]}{[\text{ACe}^{3+}3\text{d}_{5/2} + \text{ACe}^{3+}3\text{d}_{3/2} + \text{A}'\text{Ce}^{3+}3\text{d}_{5/2} + \text{A}'\text{Ce}^{3+}3\text{d}_{3/2} + \text{ACe}^{4+}3\text{d}_{5/2} + \text{ACe}^{4+}3\text{d}_{3/2}]}$$

where A is the integrated area under each deconvoluted peak.

Based on this calculation, the concentration of [Ce³⁺] and [Ce⁴⁺] was found to be 0.65 and 0.35, respectively. The O 1s

core spectrum of Dex-CeO₂ NPs was deconvoluted into two peaks at 531.6 eV and 530.50 eV, corresponding to surface oxygen and lattice oxygen, respectively (Fig. S1C†). The high-resolution C 1s spectrum was also deconvoluted into two peaks with binding energies at 284.65 eV and 285.8 eV, which are attributed to C–C and C–OH bonds, respectively (Fig. S1D†). In Dex-Mn₃O₄ NPs, the Mn 2p core spectrum was deconvoluted into two spin-orbit pairs of 2p_{3/2}/2p_{1/2} at 640.10 eV/651.93 eV and 642.86 eV/654.64 eV, indicating mixed Mn²⁺ and Mn³⁺ valence states, respectively (Fig. 1F).²⁶ The O 1s spectrum of Dex-Mn₃O₄ NPs revealed peaks at 530.48 eV and 531.85 eV, corresponding to Mn–OH and H–O–H bonds (Fig. S2C†). Additionally, the high-resolution C 1s spectrum was deconvoluted into three peaks at 284.72, 285.95, and 286.51 eV which were identified as corresponding to C–C/C=C, C–O–C, and C=O bonds, respectively (Fig. S2D†). EDX analysis of both the nanoparticles showed prominent signals for carbon (C), oxygen (O), and the respective elements, Ce or Mn, with characteristic energy lines, Ce (0.88 and 4.84 keV) and Mn (5.8 and 0.6 keV), confirming the elemental composition (Fig. S1E and S2F†). Elemental mapping of Dex-CeO₂ NPs displayed distinct signals for Ce and O, with an overlay indicating the oxide of cerium, while the overlapping signals of Mn and O suggested a strong Mn–O interaction and thus the presence of Mn oxide (Fig. S1F and S2F†).^{27,28}

The typical Fenton reaction, involving oxidation of H₂O₂ by the ferrous ions (Fe²⁺), was utilized to generate ·OH²⁹ (Fig. S3A†). The high affinity of ·OH with a specific fluorescent probe, terephthalic acid (TA), was used to probe the generation of the former. The so-generated fluorescent product, 2-hydroxyterephthalic acid, could be easily monitored by following the Ex./Em. at 320/425 nm. The reaction of FeSO₄ with H₂O₂ in the presence of TA showed the maximum emission intensity (Fig. 2A, black curve), due to the formation of 2-hydroxyterephthalic acid, suggesting the possible formation of ·OH. To this suspension, the addition of increasing concentrations of Dex-CeO₂ NPs (25–200 μM) and Dex-Mn₃O₄ NPs (25–200 μM) caused a decrease in the emission intensity of terephthalic acid (Fig. S3B and S3C†). Furthermore, the scavenging kinetics of ·OH by the nanozymes were followed for 20 minutes. The results revealed that with the increase in the concentration of the nanozymes, there was a concomitant decrease in the TA emission intensity at 425 nm (Fig. 2A and B). These results indicate that Dex-CeO₂ NPs and Dex-Mn₃O₄ NPs could effec-

tively scavenge ·OH even in the event of constant generation of ·OH, typically apparent during the biochemical reactions in mammalian cells. The possibility of TA oxidation by H₂O₂

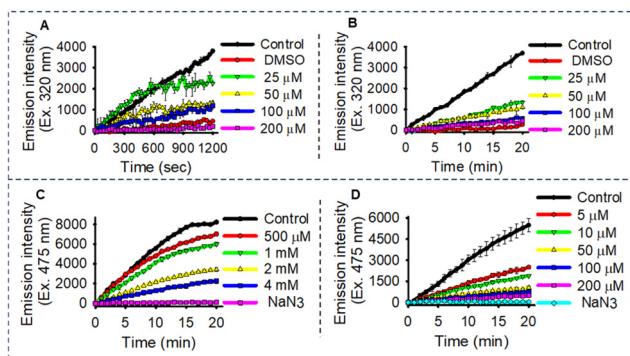


Fig. 2 Reaction kinetics of ·OH scavenging by different concentrations of (A) Dex-CeO₂ NPs and (B) Dex-Mn₃O₄ NPs with the control as FeSO₄ + H₂O₂ + TA. Scavenging of ¹O₂ by different concentrations of (C) Dex-CeO₂ NPs and (D) Dex-Mn₃O₄ NPs with the control as HRP + H₂O₂ + SOSG. All the experiments were performed at least thrice in triplicate and the data are plotted with the standard deviation.

alone (without ·OH radicals) was tested using two methods, by substituting FeSO₄ (Fe²⁺) with FeCl₃ (Fe³⁺) in the Fenton reaction system and direct exposure of TA to H₂O₂ (Fig. S4†). Both of these reactions failed to produce detectable TA oxidation, which suggests the requirement for Fe²⁺ in catalyzing ·OH generation *via* the Fenton reaction and rules out the possibility of false positive signals of TA oxidation by only H₂O₂.

A significant reduction in the emission intensity of 2-hydroxyterephthalic acid in presence of 10% DMSO (Fig. S5A and S5C†) and 10% ethanol (Fig. S5B and S5D†) relative to the control (FeSO₄ + H₂O₂ in a buffer) was observed to be analogous to 50 μM of Dex-CeO₂ NPs and Dex-Mn₃O₄ NPs.

The generation of ¹O₂ was achieved by utilizing the catalytic reaction between horseradish peroxidase (HRP) and H₂O₂ at pH 6 in the presence of SOSG (Fig. S6A†), a selective fluorescent probe for ¹O₂, due to the formation of SOSG-endoperoxide (SOSG-EP) (Ex./Em. = 475/530 nm). The reaction system used for ¹O₂ generation consisted of HRP and H₂O₂ in a buffer at pH 6 (Fig. S6A,† black curve). NaN₃ was used as a scavenger of ¹O₂. HRP, in the presence of H₂O₂, generates O₂^{·-}, which serves as a source of ¹O₂ after oxidation. In a study by Ingenbosch *et al.*, the catalytic reaction between HRP and H₂O₂ was carried out in the presence of the superoxide dismutase (SOD) enzyme, a known O₂^{·-} scavenger. Although the SOSG-EP fluorescence decreased with increasing SOD concentration, there was no complete quenching of the fluorescence, indicating the major role of O₂^{·-} in ¹O₂ formation. We also tested the hypoxanthine/xanthine oxidase system, known to generate O₂^{·-}, using SOSG at pH 6, but it did not show significant emission at 530 nm, indicating the absence of ¹O₂ (Fig. S6B,† red curve). Using DHE, a probe for O₂^{·-} detection, we observed no emission in the presence of *p*-benzoquinone (O₂^{·-} inhibitor) and no decrease in emission intensity in the presence of NaN₃, suggesting that the hypoxanthine/xanthine oxidase system only produced O₂^{·-} and no ¹O₂ (Fig. S6C†). Subsequently, we also tested the formation of O₂^{·-} in the HRP and H₂O₂ system and compared it with the hypoxanthine/

xanthine oxidase system using DHE (Fig. S6D†). DHE showed excellent emission when incubated with hypoxanthine/xanthine oxidase; however, HRP + H₂O₂ with DHE showed weak emission intensity. This observation suggests that the HRP + H₂O₂ reaction produces predominantly ¹O₂ with a minor population of O₂^{·-}, possibly contributing to the formation of ¹O₂ in the system. The ¹O₂ scavenging potential of Dex-CeO₂ NPs and Dex-Mn₃O₄ NPs was estimated by following the fluorescence emission spectra of SOSG-EP at 530 nm (Fig. S7A†). The control reaction consisting of HRP and H₂O showed strong emission intensity (Fig. S7B and S7C,† black curve). In the presence of Dex-CeO₂ NPs (500 μM–4 mM) and Dex-Mn₃O₄ NPs (5–200 μM), the emission intensity of SOSG-EP was observed to decrease in a concentration-dependent manner, which suggests the successful scavenging of ¹O₂ by the nanozymes (Fig. S7B and S7C†). A similar trend of inhibition of ¹O₂ could be seen at various concentrations of Dex-CeO₂ NPs (500 μM–4 mM) and Dex-Mn₃O₄ NPs (5–200 μM) for a 20-minute kinetics study, which further validates their scavenging ability (Fig. 2C and D). Dex-Mn₃O₄ NPs possess stronger ¹O₂ scavenging potential (~20 times) than Dex-CeO₂ NPs, as evident from their extent of scavenging ability at lower concentrations (5–200 μM). A notable reduction in the emission intensity of SOSG-EP was observed in the presence of the inhibitors, NaN₃ (Fig. S8A and S8C†) and *p*-benzoquinone (Fig. S8B and S8D†), relative to the control group (HRP and H₂O₂ in buffer). This is analogous to the inhibitory effects of Dex-CeO₂ NPs (1 mM) and Dex-Mn₃O₄ NPs (10 μM).

The possible byproducts generated by the scavenging of ·OH could be H₂O₂ and O₂. Therefore, we tested the supernatant of the Fenton reaction system, employed for studying the scavenging activity of NPs, and estimated the generation of H₂O₂ by the Amplex Red assay. The generation of molecular oxygen was estimated using an oxygen-sensitive probe. The ·OH scavenging by CeO₂ NPs neither displayed the formation of molecular oxygen nor H₂O₂ (Fig. 3A, B, and S9A†), which indicates that ·OH is being catalytically converted into H₂O possibly by the following reaction: [Ce₂O₃ (Ce³⁺) + 2(·OH) → 2CeO₂ (Ce⁴⁺) + H₂O]. A similar observation has been reported by Xue *et al.*³⁰ Thus, the observed scavenging activity of CeO₂ NPs could be well correlated with the unique ability to switch between their dual oxidation states Ce³⁺ and Ce⁴⁺. The relatively higher concentration of Ce³⁺ at the surface of CeO₂ NPs acts as the active sites for the redox reaction and allows them to react with the powerful oxidant, ·OH, by reversibly switching between the Ce³⁺ and Ce⁴⁺ ions.³¹ Interestingly, the ·OH scavenging by Dex-Mn₃O₄ NPs showed significant liberation of O₂ at higher concentrations (200 μM) (Fig. 3C) than at lower concentrations (100 μM). Additionally, the Amplex Red test showed stronger absorbance at 570 nm at both the concentrations of Dex-Mn₃O₄ NPs (100 μM and 200 μM) compared to the control (FeSO₄ and H₂O₂). These observations indicate the formation of H₂O₂ in the reaction system (Fig. 3D and S9B†). These experimental results indicate that H₂O₂ may form as a byproduct during the ·OH scavenging activity of Dex-Mn₃O₄ NPs. However, at higher concentrations of Dex-Mn₃O₄ NPs, the

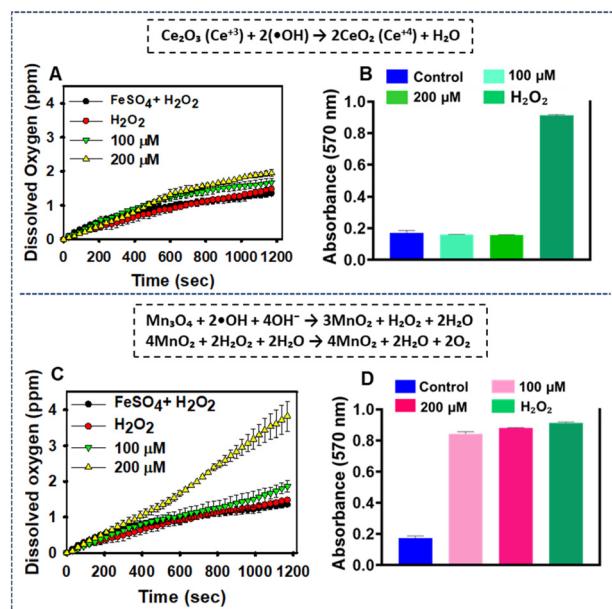
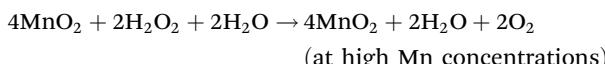
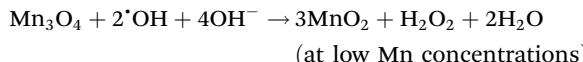


Fig. 3 Possibility of conversion of ·OH into O₂ or H₂O₂ by nanozymes: Conversion of ·OH into dissolved oxygen (A) and H₂O₂ (B) by Dex-CeO₂ NPs. Conversion of ·OH into dissolved oxygen (C) and H₂O₂ (D) by Dex-Mn₃O₄ NPs. Detection of H₂O₂ formation was performed using the Amplex Red assay whereas dissolved oxygen was measured using a DO meter. All the experiments were performed three times independently and the data are plotted with the standard deviation.

generated H₂O₂ could auto-degrade into O₂ and H₂O as evidenced by the generation of O₂ at 200 µM of Dex-Mn₃O₄ NPs (Fig. 3C). The probable reaction mechanism for the ·OH scavenging activity of Dex-Mn₃O₄ NPs can be represented as follows:

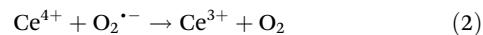
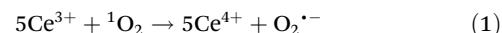


Thus, both the nanozymes follow different reaction mechanisms to scavenge ·OH at physiological pH.

Furthermore, the mechanism for ¹O₂ scavenging by Dex-CeO₂ and Dex-Mn₃O₄ NPs could possibly involve a series of redox reactions, with the formation of O₂^{·-} as an intermediate species and O₂ as the end product. During the initial steps, the nanozymes act as electron donor to ¹O₂, facilitating its reduction into O₂^{·-}.³² This step is favoured by the half-reaction: ¹O₂ + e⁻ → O₂^{·-} ($E^\circ = +0.81$ eV). In this step, Ce³⁺ atoms from CeO₂ NPs undergo redox switching to Ce⁴⁺ oxidation state as evidenced by the feasibility of the half-reaction, Ce⁴⁺ + e⁻ → Ce³⁺ ($E^\circ = +1.72$ eV).³³ During the second course of reaction, Ce⁴⁺ is reduced to Ce³⁺ by the following reaction: Ce⁴⁺ + O₂^{·-} → Ce³⁺ + O₂. Korsvik *et al.* have proposed a similar

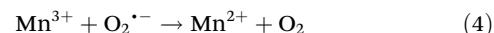
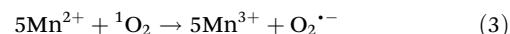
mechanism for O₂^{·-} dismutation by CeO₂ NPs, analogous to the activities of Fe- and Mn-SODs.²⁵

The overall redox reactions involving the ¹O₂ scavenging mechanism of Dex-CeO₂ NPs can be summarized as:



In the case of Mn₃O₄ NPs, the Mn²⁺ ions donate electrons to reduce ¹O₂ to O₂^{·-} (Mn²⁺ → Mn³⁺ + e⁻ ($E^\circ = +1.509$ eV)). Subsequently, the generated O₂^{·-} is converted back into O₂ via oxidation by the following reaction: O₂^{·-} → O₂ + e⁻ ($E^\circ = +0.33$ eV). This step involves electron transfer between O₂^{·-} and the nanozyme, resulting in the reduction of the nanozyme and regeneration of their oxidized state. Mn³⁺ is reduced back to Mn²⁺ by the following reaction: Mn³⁺ + e⁻ → Mn²⁺ ($E^\circ = +1.51$ eV). The overall scavenging activity of the nanoparticle involves the reduction of ¹O₂ to O₂^{·-}, followed by the conversion of O₂^{·-} to O₂, thereby effectively neutralizing ¹O₂.

The mechanism of ¹O₂ scavenging by Dex-Mn₃O₄ NPs could be possibly written as:



Learman *et al.* have experimentally demonstrated the role of O₂^{·-} as an effective oxidant of Mn²⁺, promoting the formation of Mn³⁺ oxide. Mechanistically, the reaction was thought to proceed through the formation of a Mn³⁺ intermediate, thereby facilitating the oxidation of Mn²⁺ with concomitant scavenging of ¹O₂.³⁴

The IC₅₀ values, representing the half-maximal inhibitory concentration, were determined for scavenging of both ·OH and ¹O₂ by Dex-CeO₂ NPs (Fig. S10A, B and Fig S11A, B†) and Dex-Mn₃O₄ NPs (Fig. S10C, D and Fig S11C, D†) and compared with those of known scavengers, NAC for ·OH (Fig. S10E and F†) and NaN₃ for ¹O₂ (Fig. S11E and F†). The results revealed that Dex-Mn₃O₄ NPs exhibited the best scavenging efficiency for both ·OH [2.3 µM] and ¹O₂ [4.75 µM] compared to the well-known scavengers NAC [28.4 µM] and NaN₃ [60.5 µM] (Table S2†).

Furthermore, Dex-CeO₂ NPs also showed comparable scavenging of ·OH [5.4 µM] with those of Dex-Mn₃O₄ NPs [2.3 µM] and NAC [28.4 µM]; however, poor scavenging of ¹O₂ [857.27 µM] compared to Dex-Mn₃O₄ NPs [4.75 µM] and NaN₃ [60.5 µM] was observed. Specifically, Dex-Mn₃O₄ NPs exhibited significantly lower IC₅₀ values for both the radicals, indicating superior scavenging capabilities. Furthermore, Dex-CeO₂ NPs outperformed NAC in scavenging ·OH; however, they were less efficient than Dex-Mn₃O₄ NPs.

CeO₂ NPs are well known for their ability to undergo reversible redox cycling between the Ce³⁺ and Ce⁴⁺ oxidation states, which is the mechanistic basis for their enzyme-mimetic functions.³⁵ We also tested the redox cycling behavior of Dex-CeO₂ NPs (Fig. S12H†) and compared that with bare CeO₂ NPs (Fig. S12G†). The colourless suspensions of bare CeO₂ NPs (Fig. S12G,† bottle 1) and Dex-CeO₂ NPs (Fig. S12H,† bottle 1)

developed into dark yellow suspensions immediately after H_2O_2 addition, indicating the oxidation of Ce^{3+} to Ce^{4+} (Fig. S12G,† bottle 2 and Fig. S12H,† bottle 2). Following incubation at 37 °C for 15 days, the suspensions reverted to the colourless state, confirming the regeneration of Ce^{3+} during the redox transition (Fig. S12G,† bottle 3 and Fig. S12H,† bottle 3). To evaluate the persistence of redox reversibility, H_2O_2 was again introduced into the regenerated colourless suspensions. A progressive development of the yellow colouration was again observed (Fig. S12G,† bottle 4 and Fig. S12H,† bottle 4), indicating a second oxidation cycle of conversion of Ce^{3+} into Ce^{4+} . This reversible colour change upon repeated H_2O_2 exposure provides strong evidence of sustained redox cycling by Dex- CeO_2 NPs under physiological conditions. We also investigated if the redox cycling of Dex- CeO_2 NPs affects their ability to scavenge $\cdot\text{OH}$ and $\cdot\text{O}_2$ (Fig. S12A, B, C and D†). The $\cdot\text{OH}$ and $\cdot\text{O}_2$ scavenging activity of Dex- CeO_2 NPs post-redox recycling showed no significant deviation in comparison with the respective controls. Results from UV-vis absorbance spectroscopy further support this observation as there was no appreciable change in the absorbance signal of bare CeO_2 NPs at 280–290 nm (Fig. S12E†) and Dex- CeO_2 NPs at 290 nm (Fig. S12F†) following redox cycling, indicating that the optical properties of the Dex- CeO_2 NPs remain unaltered throughout the process.

The *in vitro* biocompatibility assessment of Dex- CeO_2 NPs and Dex- Mn_3O_4 NPs was performed in intestinal epithelial (IEC-6) cells by incubating for 24, 48, and 72 hours with different concentrations (100 ng mL^{-1} – $1 \mu\text{g mL}^{-1}$). The nanozymes demonstrated excellent stability in serum-free and serum-containing cell culture media (DMEM-F12). The nanozymes neither showed aggregation nor any significant decrease in $\cdot\text{OH}$ and $\cdot\text{O}_2$ radical scavenging ability (Fig. S13A, B, C, D, E and F†). These results confirm the stability of dextran-coated nanozymes in physiologically relevant media. The MTT assay results showed no significant reduction in cell viability after exposure to Dex- CeO_2 NPs (Fig S14A†) and Dex- Mn_3O_4 NPs (Fig S14D†) for 24 and 48 hours. However, higher concentrations of Dex- Mn_3O_4 NPs ($0.75 \mu\text{g mL}^{-1}$ and $1 \mu\text{g mL}^{-1}$) caused an ~10–15% decrease in cell viability when incubated for 72 hours. This observation could be due to the deposition of Dex- Mn_3O_4 NPs (higher concentrations) on the cells, causing physical damage. The cell morphology revealed no significant alteration in the growth and adhesion pattern of typical IEC-6 cells when exposed to different concentrations of Dex- CeO_2 NPs (Fig. S15†) and Dex- Mn_3O_4 NPs (Fig. S16†). The cell cycle pattern analysis in a large population of IEC-6 cells exposed to Dex- CeO_2 NPs and Dex- Mn_3O_4 NPs indicated no significant alteration in the distribution of cell cycle phases (G1, S, and G2/M) compared to the control cells (Fig S14B and S14E†). Quantitative analysis revealed that the proportion of cells in each phase (G1, S, and G2/M) of the treated groups remained comparable to that of the control cells. This suggests that treatment with Dex- CeO_2 NPs (Fig S14C†) and Dex- Mn_3O_4 NPs (Fig S14F†) did not induce any notable changes in cell cycle progression or arrest at the tested concentrations. Furthermore,

H_2DCFDA dye-based investigation of ROS generation by higher concentrations of Dex- CeO_2 NPs and Dex- Mn_3O_4 NPs was performed. Fluorescence microscopy imaging showed strong green fluorescence signals from the positive control (cells exposed to H_2O_2) (Fig. S14G†) compared to the untreated (control) cells (Fig. S14J†) and the Dex- CeO_2 NP- (Fig S14M-O†) and Dex- Mn_3O_4 NP (Fig. S14P-R†)-treated cells. Thus, from the *in vitro* cellular experiments, it may be concluded that the developed antioxidant nanozymes are biocompatible to mammalian cells and do not elicit any oxidative stress and toxicity even at high concentrations.

In conclusion, the developed nanozymes efficiently scavenge $\cdot\text{OH}$ and $\cdot\text{O}_2$ in a concentration- and time-dependent manner. Dex- Mn_3O_4 NPs exhibited superior scavenging of $\cdot\text{OH}$ and $\cdot\text{O}_2$ compared to Dex- CeO_2 NPs and other specific scavengers, NAC and NaN_3 . Mechanistically, Dex- CeO_2 NPs neutralize $\cdot\text{OH}$ directly to produce water, while Dex- Mn_3O_4 NPs convert $\cdot\text{OH}$ into H_2O_2 , subsequently breaking it into O_2 . Additionally, both the nanozymes effectively neutralize $\cdot\text{O}_2$ by converting it into O_2 . The developed nanozymes were found to be biocompatible as their exposure did not affect the cell viability and induce oxidative stress, and the cell cycle progression pattern was also unaffected. Considering the growing interest in utilizing nanozymes for clinical applications, particularly in the treatment of oxidative-stress-associated diseases, both CeO_2 - and Mn_3O_4 -based nanozymes would be promising candidates for therapeutic applications. Although the results from *in vitro* experiments indicate that the developed nanozymes are non-toxic to mammalian cells, the *in vivo* safety assessment remains to be studied in detail before these nanozymes are considered for clinical studies.

Data availability

The data supporting this article have been included as part of the ESI.†

Conflicts of interest

There are no conflicts to declare.

Acknowledgements

S. Singh thanks the NIAB Core Grant (C0046) for financial support. K. M. R. thanks the University Grant Commission (UGC) for providing a Junior Research Fellowship. D. Mehta thanks the Department of Biotechnology (DBT), India, for providing a Senior Research Fellowship.

References

- 1 K. Murotomi, A. Umeno, M. Shichiri, M. Tanito and Y. Yoshida, *Int. J. Mol. Sci.*, 2023, **24**, 2739.

2 G. Cui, S. Dong, C. Sui, T. Kakuchi, Q. Duan and B. Feng, *Int. J. Polym. Mater. Polym. Biomater.*, 2022, **71**, 109–115.

3 S. Yang, Y. Liu, T. Wu, X. Zhang, S. Xu, Q. Pan, L. Zhu, P. Zheng, D. Qiao and W. Zhu, *J. Med. Chem.*, 2025, **1**.

4 Q. Dong and Z. Jiang, *Inorganics*, 2024, **12**, 331.

5 Y. Chen, Y. Deng, Y. Li, Y. Qin, Z. Zhou, H. Yang and Y. Sun, *ACS Appl. Mater. Interfaces*, 2024, **16**, 21546–21556.

6 C. Feng, Y. Wang, J. Xu, Y. Zheng, W. Zhou, Y. Wang and C. Luo, *Pharmaceutics*, 2024, **16**, 1582.

7 N. K. Panchal and E. P. Sabina, *Food Chem. Toxicol.*, 2023, **172**, 113598.

8 T. Pirmohamed, J. M. Dowding, S. Singh, B. Wasserman, E. Heckert, A. S. Karakoti, J. E. King, S. Seal and W. T. Self, *Chem. Commun.*, 2010, **46**, 2736–2738.

9 S. Singh, T. Dosani, A. S. Karakoti, A. Kumar, S. Seal and W. T. Self, *Biomaterials*, 2011, **32**, 6745–6753.

10 L.-Y. Tsai, K.-T. Lee and T.-Z. Liu, *Free Radicals Biol. Med.*, 1998, **24**, 732–737.

11 M. B. Bogdanov, L. E. Ramos, Z. Xu and M. F. Beal, *J. Neurochem.*, 1998, **71**, 1321–1324.

12 H.-A. Arab, R. Jamshidi, A. Rassouli, G. Shams and M. Hassanzadeh, *Brit. Poult. Sci.*, 2006, **47**, 216–222.

13 D. A. Parks and D. N. Granger, *Am. J. Physiol.: Gastrointest. Liver Physiol.*, 1983, **245**, G285–G289.

14 V. Vanasco, P. Evelson, A. Boveris and S. Alvarez, *Chem.-Biol. Interact.*, 2010, **184**, 313–318.

15 K. Murotomi, A. Umeno, M. Yasunaga, M. Shichiri, N. Ishida, H. Abe, Y. Yoshida and Y. Nakajima, *Free Radical Res.*, 2015, **49**, 133–138.

16 A. Filippi, F. Liu, J. Wilson, S. Lelieveld, K. Korschelt, T. Wang, Y. Wang, T. Reich, U. Pöschl and W. Tremel, *RSC Adv.*, 2019, **9**, 11077–11081.

17 Y. Ogawa, T. Kawaguchi, M. Tanaka, A. Hashimoto, K. Fukui, N. Uekawa, T. Ozawa, T. Kamachi and M. Kohno, *J. Clin. Biochem. Nutr.*, 2023, **73**, 1.

18 T. O. Shekunova, L. A. Lapkina, A. B. Shcherbakov, I. N. Meshkov, V. K. Ivanov, A. Y. Tsivadze and Y. G. Gorbunova, *J. Photochem. Photobiol. A*, 2019, **382**, 111925.

19 M. Bancirova, *Luminescence*, 2011, **26**, 685–688.

20 K. C. Das and H. P. Misra, *Mol. Cell. Biochem.*, 2004, **262**, 127–133.

21 D. Mehta, P. Sharma and S. Singh, *Colloids Surf. B*, 2023, **231**, 113531.

22 R. Regmi, R. Tackett and G. Lawes, *J. Magn. Magn. Mater.*, 2009, **321**, 2296–2299.

23 G. Wang, Q. Mu, T. Chen and Y. Wang, *J. Alloys Compd.*, 2010, **493**, 202–207.

24 S. Deshpande, S. Patil, S. V. Kuchibhatla and S. Seal, *Appl. Phys. Lett.*, 2005, **87**, 2.

25 C. Korsvik, S. Patil, S. Seal and W. T. Self, *Chem. Commun.*, 2007, 1056–1058.

26 J. Duan, S. Chen, S. Dai and S. Z. Qiao, *Adv. Funct. Mater.*, 2014, **24**, 2072–2078.

27 N. Yadav and S. Singh, *Emergent Mater.*, 2021, 1–13.

28 A.-N. Chowdhury, M. S. Azam, M. Aktaruzzaman and A. Rahim, *J. Hazard. Mater.*, 2009, **172**, 1229–1235.

29 F. Ducrozet, A. Sebastian, C. J. G. Villavicencio, S. Ptasinska and C. Sicard-Roselli, *Phys. Chem. Chem. Phys.*, 2024, **26**, 8651–8657.

30 Y. Xue, Q. Luan, D. Yang, X. Yao and K. Zhou, *J. Phys. Chem. C*, 2011, **115**, 4433–4438.

31 S. Duanghathaipornsk, F. A. Alateeq, S. S. Kim, D.-S. Kim and A. C. Alba-Rubio, *Sens. Actuators, B*, 2020, **321**, 128467.

32 H. Tamura and H. Ishikita, *J. Phys. Chem. A*, 2020, **124**, 5081–5088.

33 C. Xu and X. Qu, *NPG Asia Mater.*, 2014, **6**, e90.

34 D. R. Learman, B. M. Voelker, A. S. Madden and C. M. Hansel, *Front. Microbiol.*, 2013, **4**, 262.

35 R. Singh and S. Singh, *Colloids Surf. B*, 2015, **132**, 78–84.