PARP-1 Inhibits Glycolysis in Ischemic Kidneys

Kishor Devalaraja-Narashimha* and Babu J. Padanilam*†

*Department of Cellular and Integrative Physiology and †Department of Internal Medicine, Section of Nephrology, University of Nebraska Medical Center, Omaha, Nebraska

ABSTRACT

After ischemic renal injury (IRI), selective damage occurs in the S3 segments of the proximal tubules as a result of inhibition of glycolysis, but the mechanism of this inhibition is unknown. We previously reported that inhibition of poly(ADP-ribose) polymerase-1 (PARP-1) activity protects against ischemia-induced necrosis in proximal tubules by preserving ATP levels. Here, we tested whether PARP-1 activation in proximal tubules after IRI leads to poly(ADP-ribosyl)ation of the key glycolytic enzyme glyceraldehyde-3-phosphate dehydrogenase (GAPDH), a modification that inhibits its activity. Using in vitro and in vivo models, under hypoxic conditions, we detected poly(ADP-ribosyl)ation and reduced activity of GAPDH; inhibition of PARP-1 activity restored GAPDH activity and ATP levels. Inhibition of GAPDH with iodoacetate exacerbated ATP depletion, cytotoxicity, and necrotic cell death of LLCPK1 cells subjected to hypoxic conditions, whereas inhibition of PARP-1 activity was cytoprotective. In conclusion, these data indicate that poly(ADP-ribosyl)ation of GAPDH and the subsequent inhibition of anaerobic respiration exacerbate ATP depletion selectively in the proximal tubule after IRI.


Compromised perfusion of renal tissues leading to ischemic renal injury (IRI), generally accepted as the major cause of acute kidney injury (AKI), usually results from hypoxia-induced renal vascular and tubular dysfunction. The outer medullary region of the kidney receives <10% of the blood delivered to the kidney via the renal artery. After IRI, a persistent perfusion deficit exists even at 24 h after reperfusion, and the outer medullary partial pressure of oxygen is restored to only 10% of its normal levels, rendering this region susceptible to injury at both the tubular and vascular levels.1–3 Thus, the prolonged perfusion deficit shuts down oxidative phosphorylation in the cells of the outer medullary segments of the nephron and reverts to anaerobic metabolism for ATP synthesis. Nevertheless, ischemia causes selective injury to the outer medullary proximal straight tubules (PST), causing the PST cells to undergo cell death and/or sublethal injury to instigate renal dysfunction.4,5 The medullary thick ascending limb, although situated in the same region, does not undergo injury to the same level.6–8 Despite ongoing debate for more than two decades, the molecular mechanisms by which PST cells undergo selective injury are not known.

Hypoxia resulting from decreased blood flow leads to a variety of secondary effects, including a breakdown in cellular energy metabolism and generation of reactive oxygen species (ROS) and reactive nitrogen species.9,10 The superoxides induce DNA strand breaks in ischemic kidneys as early as 1 h after IRI.11 The severe DNA damage that ensues results in excessive activation of the DNA repair enzyme poly(ADP-ribose) polymerase-1 (PARP-1), which exacerbates ATP depletion and triggers signaling cascades, leading to cellular suicide.12 Recent data from our laboratory showed selective

Received March 26, 2008. Accepted August 21, 2008.
Published online ahead of print. Publication date available at www.jasn.org.

Correspondence: Dr. Babu J. Padanilam, Department of Cellular and Integrative Physiology, 985850 Nebraska Medical Center, Omaha, NE 68198-5850. Phone: 402-559-3575; Fax: 402-559-4438; E-mail: bpadanilam@unmc.edu

Copyright © 2009 by the American Society of Nephrology
upregulation of PARP-1 expression and its activity in PST cells after renal ischemia.13-14 Gene ablation or pharmacologic inhibition of PARP-1 activity offers both functional and histopathologic protection from IRI.13,15 The ATP levels are significantly preserved in both in vivo and in vitro models of IRI after PARP gene ablation or inhibition, respectively13,15,16; however, the exact mechanisms by which PARP activation leads to ATP depletion and whether these mechanisms are linked to selective damage to PST after renal ischemia are not defined.

According to the “cell suicide” hypothesis, PARP-1 activation induces energy failure by depleting NAD⁺, and the cell consumes ATP to replete the NAD⁺ level, ultimately leading to energy failure and cell death;12 however, the role of NAD⁺ in energy depletion is controversial, and its depletion alone may not be lethal to cells.17,18 Moreover, PST cells can carry out anaerobic respiration under hypoxic conditions and be protected from injury. Nevertheless, under anoxic conditions, significant amounts of ATP are generated by anaerobic glycolysis by thick ascending limbs but not by the proximal tubular cells.19 These findings prompted us to investigate whether PARP activation interferes with glycolytic ATP synthesis and thus exacerbates ATP depletion and PST cell injury.

Glyceraldehyde-3-phosphate dehydrogenase (GAPDH) is a key enzyme in the glycolytic pathway and is susceptible to several modifications that alter its activity, including oxidative modification of thiols and mono-ADP-ribosylation.20-22 Recently, PARP-1 was reported to inhibit GAPDH activity by poly(ADP-ribosyl)ation after hyperglycemia-induced aortic endothelial cell injury.23 The temporal and spatial expression pattern of PARP-1 in PST during the time of ischemic injury prompted us to hypothesize that poly(ADP-ribosyl)ation and inhibition of the GAPDH leads to inhibition of glycolysis, reducing ATP synthesis and exacerbating energy depletion and cell injury.

In this study, we evaluated the role of GAPDH-poly(ADP-ribosyl)ation as a mechanism to inhibit GAPDH activity in PST after IRI using in vitro and in vivo models. We explored the role of GAPDH-poly(ADP-ribosyl)ation in inhibiting glycolysis, exacerbating ATP depletion, and inducing cell death. Our findings suggest that PARP-1-mediated anaerobic glycolytic inhibition is a key mechanism of selective PST injury after IRI.

RESULTS

GAPDH Activity Assay in Wild-Type and PARP⁻/⁻ Mouse Kidneys after IRI

To test our hypothesis that GAPDH activity is inhibited by PARP, we first evaluated GAPDH activity in wild-type (WT)
and PARP−/− renal outer medullary proteins after IRI. As shown in Figure 1, GAPDH activity was significantly inhibited at 3 and 6 h after IRI in WT mouse kidneys compared with that of sham-operated kidneys, whereas its activity was significantly improved at both 3 and 6 h after IRI in PARP−/− mouse kidneys compared with their WT counterparts. These data suggest a role for PARP-1 in the inhibition of GAPDH activity after IRI. No significant change in GAPDH activity was observed between WT and PARP−/− sham-operated kidneys.

GAPDH-Poly(ADP-ribosyl)ation in Ischemic Renal Tissues

Because activated PARP-1 ADP ribosylates several different proteins and modulates their activities, we next examined whether poly(ADP-ribosyl)ation of GAPDH could be a possible underlying mechanism for its reduced activity after IRI. Proteins isolated from the outer medullary region of WT or PARP−/− animals that underwent either sham surgery or IRI were subjected to immunoblot analysis. Several poly(ADP-ribosyl)ated protein bands, including a band at approximately 37 kD, were observed at 3 and 6 h after IRI in WT kidney lysates (Figure 2A, lanes 1 and 2, respectively) with anti-PAR antibody, suggesting possible GAPDH-poly(ADP-ribosyl)ation after IRI. No PAR bands were observed in either sham-operated or PARP−/− ischemic renal tissues (data not shown).

Lactate and ATP levels in WT and PARP−/− Mouse Kidneys after IRI

To show that the inhibition of GAPDH activity by PARP-1 does indeed inhibit anaerobic glycolysis, we measured lactate production and ATP levels in the outer medulla of WT and PARP−/− mouse kidneys after IRI. As shown in Figure 3A, lactate production was significantly higher in the outer medulla of PARP−/− mice compared with WT mice at 12 h after IRI. As shown in Figure 3B, the ATP levels in the outer medulla of WT mice at 12 h after IRI were decreased to 58.5 ± 8.0% of the sham-operated mice (P < 0.03; n = 4), whereas the absence of PARP-1 preserved ATP levels to 91.5 ± 9.2% of sham-operated mice. The ATP levels in sham-operated kidneys were measured to be 11.14 ± 0.6 nmol/mg protein.

Effect of PARP Inhibition on GAPDH Activity in Hypoxia-Injured LLCPK1 Cells

To confirm our in vivo finding that PARP-1 mediates the inhibition of GAPDH activity and to demonstrate that this mechanism occurs in PST cells, we used the porcine-derived proximal tubular cell line LLCPK1. LLCPK1 cells expressed proximal tubular marker aquaporin 1 (AQP1) and the PST marker AQP7 (see Supplementary Figure S1A). Furthermore, 6 h of
Inhibition of GAPDH Activity by PARP-1 Is Due to its Poly(ADP-ribosyl)ation

To confirm further the direct role of PARP-1 in the inhibition of GAPDH activity, we performed an in vitro cell-free assay involving purified PARP-1 and GAPDH proteins. GAPDH activity was significantly inhibited by activated PARP-1 (PARP-1 + activated DNA) in the presence of NAD\(^+\) compared with GAPDH alone (Figure 5, bar 3 versus bar 1). Presence of the PARP-1 inhibitor GPI-15427 reversed the effect of activated PARP-1 on GAPDH activity (Figure 5, bars 4 and 5 versus bar 3), indicating a direct role of PARP-1 activity in the inhibition of GAPDH activity. Inclusion of NAD\(^+\) alone in the incubation medium, without activated PARP-1, did not change GAPDH activity significantly (Figure 5, bar 2 versus bar 1). The proteins from the in vitro cell-free assay, when subjected to immunoblotting with anti-PAR antibody, demonstrated PAR modification of PARP and GAPDH in a PARP-1–dependent manner (see Supplementary Figure S2, B through E).

Effect of Hypoxia on ATP Levels in Cultured LLCPK\(_1\) Cells

To examine whether energy levels are compromised after hypoxic injury in LLCPK\(_1\), and whether PARP inhibitor preserves them, we measured ATP levels in LLCPK\(_1\) cells immediately after hypoxic treatment so as to isolate ATP production by anaerobic respiration from that of aerobic respiration. As a positive control to indicate the significance of GAPDH activity in anaerobic ATP production, we used a GAPDH inhibitor, sodium iodoacetate (IA), as described previously.\(^{26,27}\) A 45-min incubation of LLCPK\(_1\) cells with IA resulted in significant inhibition of GAPDH activity compared with control. The inhibitory effect of IA was dosage-dependently enhanced with the increased concentration of the inhibitor (see Supplementary Figure S3). The concentration of iodoacetamide that was used in the study is comparable or lesser than the concentrations used in previous reports.\(^{26,28,29}\) ATP levels were significantly decreased in LLCPK\(_1\) cells after 6 h of hypoxic treatment when compared with control treatment (Figure 6, bar 3 versus bar 1). The PARP inhibitor GPI-15427 significantly preserved ATP levels in 6-h hypoxic-LLCPK\(_1\) cells compared with hypoxia only (Figure 6, bar 4 versus bar 3). Interestingly, inhibiting GAPDH enzyme activity by IA along with 6 h of hypoxic treatment further decreased ATP levels significantly compared with hypoxia only, and this decrease was enhanced with the increasing concentration of GAPDH inhibitor (Figure 6, bars 6 through 8 versus bar 3), suggesting the significance of preserving GAPDH activity and ATP production from anaerobic glycolytic pathway. A concentration of 1 \(\mu\)M IA did not significantly decrease ATP levels compared with hypoxia only (Figure 6, bar 5 versus bar 3). Inhibition of PARP activity in control LLCPK\(_1\) cells did not cause significant change in ATP levels (Figure 6, bar 2 versus bar 1).

Effect of GAPDH/PARP Inhibition on Cytotoxicity in Hypoxic LLCPK\(_1\) Cells

We then examined the percentage of cytotoxicity in hypoxic LLCPK\(_1\) cells by measuring the concentration of lactate dehydrogenase (LDH) that was released from injured cells. We sub-
jected the LLCPK<sub>1</sub> cells to hypoxia with and without reoxygenation and in the presence/absence of GAPDH or PARP inhibitors and measured the percentage of cytotoxicity. The cytotoxicity percentage was significantly increased when GAPDH inhibitor was used along with 6 h of hypoxia compared with hypoxia only treatment (Figure 7A, bar 2 versus bar 1), whereas PARP inhibition significantly protected cells from injury after 6 h of hypoxia (Figure 7A, bar 3 versus bar 1). Similar results were found when LLCPK<sub>1</sub> cells were subjected to 6 h of hypoxia followed by 6 h of reoxygenation; GAPDH inhibition significantly increased cytotoxicity, whereas PARP inhibition protected cells (Figure 7B).

Our results from LDH assay experiments were further bolstered by Trypan blue spectrophotometric assay for necrotic cell death. We subjected LLCPK<sub>1</sub> cells to 6 h of hypoxia followed by 6 h of reoxygenation and measured Trypan blue absorbance, which was directly proportional to necrotic cell death. As in the case of LDH assay experiments, GAPDH inhibition together with hypoxia/reoxygenation significantly increased cell death compared with hypoxia/reoxygenation only (Figure 8, bar 2 versus bar 1), whereas PARP inhibition significantly decreased cell death (Figure 8, bar 3 versus bar 1). The images in Figure 8 represent Trypan blue–stained cells corresponding to the respective treatments. Trypan blue staining seemed to be enhanced when GAPDH inhibitor was used along with hypoxia/reoxygenation compared with hypoxia/reoxygenation only. Only trace levels of Trypan blue staining were visible in the hypoxic cells treated with the PARP inhibitor GPI-15427. Collectively, these data clearly indicate that PARP-mediated GAPDH inhibition after simulated ischemia led to exacerbation of cell injury in LLCPK<sub>1</sub> cells.

**DISCUSSION**

The S3 segment of the proximal tubule or PST is extremely susceptible to IRI compared with other segments of the nephron<sup>4,5</sup>; however, the S3 segment is less vulnerable to injury compared with S<sub>1</sub> and S<sub>2</sub> segments after selective inhibition of glycolytic process or mitochondrial respiration as opposed to IRI.<sup>30</sup> Although altered hemodynamic factors and differential glycolytic enzyme concentrations or activities were proposed to be the underlying causes,<sup>1,31–33</sup> the exact mechanisms leading to selective PST injury are largely unknown.

Our data clearly demonstrated that GAPDH activity is improved in the absence of PARP-1 compared with its presence after renal ischemia in the outer medullary nephron segments. We showed that GAPDH is poly(ADP-ribosyl)ated after IRI, suggesting the reduced GAPDH activity is due to its modification by poly(ADP-ribosyl)ation. Our previous finding that PARP-1 expression and activity are selectively upregulated in the S<sub>3</sub> segments<sup>1,14</sup> but not in other tubular segments after renal ischemia suggests that GAPDH-poly(ADP-ribosyl)ation and consequent inhibition of its activity exclusively occur in
the S3 segment. Thus, although significant, the relatively small absolute difference that is observed in GAPDH activity (Figure 1) may be due to the relative changes in its activity in the whole outer medullary nephron segments used in the assay. GAPDH expression level was not altered among sham, WT, or PARP−/− ischemic renal tissues, thereby excluding the possibility of reduced expression as a mechanism of altered activity. Although we did not examine the role of ROS in GAPDH activity inhibition in this study, our previous studies did not observe any changes in the generation of ROS between WT and PARP−/− ischemic renal tissues. Moreover, data from other laboratories suggested that oxidative modification is not responsible for the reduced GAPDH activity.

To define further the mechanism and the significance of PARP-1–mediated inhibition of anaerobic ATP synthesis in exacerbation of hypoxic cell injury, we used LLCPK1 cells as an in vitro cell culture model of IRI. It has been shown that, under in vitro cell culture conditions, LLCPK1 cells tend to depend mainly on glycolysis. Similarly, under control conditions, the freshly isolated proximal straight tubular cells have higher glycolytic rates compared with proximal convoluted tubules. Hence, proximal straight tubular cells can revert to anaerobic glycolysis and be protected under hypoxia. Our data also show that lactate and ATP levels were significantly increased in the outer medulla of PARP−/− mouse kidneys compared with WT. This suggests that, under ischemia/hypoxic conditions, the glycolytic capacity of proximal straight tubular cells can be enhanced in the absence of PARP-1. We acknowledge that, although we cannot compare the glycolytic capacities of PST cells and LLCPK1 cells, our data suggest that the glycolytic capacity is increased in PST cells under hypoxia and thus the use of LLCPK1 cells as a model system can be justified. We conducted most of our in vitro experiments immediately after subjecting LLCPK1 cells to hypoxia to isolate anaerobic respiration from aerobic metabolism as the sole source of ATP production. Our data clearly demonstrated that GAPDH activity was significantly blunted in LLCPK1 cells subjected to 6 or 12 h of hypoxia, whereas PARP inhibition restored its activity after 6 h of hypoxic treatment. PARP inhibition could not restore GAPDH activity after 12 h of hypoxia, suggesting that either PARP-independent inhibition of GAPDH activity occurred under severe hypoxia or the injury at 12 h was too severe for recovery of cellular functions. Although GAPDH-poly(ADP-riboseyl)ation has been suggested to be the underlying mechanism for its inhibited activity in hyperglycemic aortic endothelial cells and in both our in vivo and in vitro models of ischemia, direct evidence is lacking toward this end. To confirm this proposed mechanism, we performed in vitro cell-free GAPDH assay using purified GAPDH and PARP-1 enzymes. We showed that GAPDH activity was significantly inhibited in the presence of NAD+ and activated PARP-1. That GAPDH activity was restored to control levels in the presence of PARP inhibitor clearly indicates that GAPDH modification by poly(ADP-riboseyl)ation leads to inhibition of its activity. Furthermore, it rules out the possibility of direct binding of PARP-1 to GAPDH as an alternate mechanism of inhibition. Western blot analysis of these proteins at the end of the in vitro cell-free GAPDH assay experiment revealed that both PARP-1 and GAPDH were poly(ADP-riboseyl)ated. To rule out the possibility that the reduction in GAPDH activity is due to nonspecific leakage of GAPDH out of the cytosol, possibly as a result of membrane damage, we assessed the levels of GAPDH protein and its activity after 6 h of hypoxia with or without PARP inhibitor in the cultured medium. Unlike the changes in the level of LDH, we observed no changes in the protein levels or activity of GAPDH (data not shown).

Under hypoxic/ischemic environment, cells cannot carry out oxidative phosphorylation because of unavailability of oxygen and hence revert to anaerobic metabolism. Measurement of ATP levels immediately after hypoxia shows the level of activation of anaerobic respiration. Our data demonstrate that ATP levels were depleted in hypoxic-LLCPK1 cells in concordance with ATP depletion, suggesting that inhibiting GAPDH activity prevents anaerobic glycolysis and consequent ATP production. We and others previously showed that PARP-1 mediates ATP depletion and subsequent necrotic cell death after simulated ischemia; however, the mechanisms by which PARP-1 elicits ATP depletion and exacerbation of PST injury after simulated ischemia was not elucidated. In this study, we showed that necrotic cell death was significantly enhanced in LLCPK1 cells subjected to hypoxia with or without reoxygenation in a PARP-1–dependent manner; however, we did not observe any apoptotic cells immediately after hypoxia (data not shown). PARP inhibition offered significant protection whereas inhibition of GAPDH enhanced necrotic cell death in hypoxic LLCPK1, in concordance with ATP depletion, suggesting GAPDH is a limiting factor in mediating anaerobic ATP synthesis and subsequent cell survival.

In summary, the study provides novel insights into the mechanisms by which PARP-1 mediates targeted acute PST...
cell injury under ischemia/hypoxia. We showed that GAPDH was poly(ADP-ribose)ylated and its activity was inhibited in both in vivo and in vitro models of simulated renal ischemia. Inhibition of GAPDH activity by PARP-1 led to decreased ATP production by anaerobic glycolysis. PARP-1–mediated inhibition of anaerobic ATP synthesis resulted in exacerbation of proximal tubular cell injury and consequent necrosis. Taken together, our data indicate that GAPDH-poly(ADP-ribose)ylation and subsequent inhibition of anaerobic respiration lead to exacerbation of ATP depletion in PST to induce selective injury after IRI.

CONCISE METHODS

Materials
All chemicals were purchased from Sigma-Aldrich Corp. (St. Louis, MO) unless otherwise noted.

Animal Models, Surgical Procedures, and Tissue Preparation
All animal procedures were performed after previous approval by the Institutional Animal Care and Use Committee at the University of Nebraska Medical Center. A total of 129 SV (WT) or PARP-1−/− mice (approximately 20 g) were purchased from Jackson Laboratories (Bar Harbor, ME). IRI was induced by bilateral renal pedicle clamping, as described previously.13 Briefly, we clamped both renal pedicles for 37 min by using microaneurysm clamps and observed reflow visually after the clamps were removed. The mice were allowed to recover for variable duration before being killed. Sham-operated mice underwent the same surgical procedure except the renal pedicles were not occluded. All mice were given free access to food and water. At the end of the each experiment, we obtained tissue from the outer medullary region (rich in PST/S3 segments) from slices made using a Stadie-Riggs microtome (Thomas Scientific, Swedesboro, NJ),36 snap-frozen with liquid nitrogen, and stored at −80°C for future experiments.

Cell Culture Conditions
The porcine-derived proximal tubular cell line LLCPK1 (ATCC, Rockville, MD) was cultured to 80 to 90% confluent monolayer cultures as described previously.39 We used hypoxia to simulate ischemic injury in in vitro experiments using a pug system providing a CO2-enriched anaerobic environment (BBL GasPak Pouch System; Becton Dickinson, Sparks, MD) as described previously.39 Samples were viewed using a Leica DMR fluorescence microscope (Thomas Scientific, Swedesboro, NJ),38 snap-frozen with liquid nitrogen, and stored at −80°C for future experiments.

GAPDH Activity Assay
We centrifuged protein lysates from frozen kidney tissue or cultured LLCPK1 cells at 4°C for 10 min at 13,000 rpm to remove cellular debris. We prepared the cytosolic fraction by centrifuging the tissue or cell lysate at 100,000 × g at 4°C for 30 min. We measured protein concentration with Bio-Rad D, protein assay reagents using the manufacturer’s protocol. We determined the GAPDH enzyme activity by the reduction of NAD+ with GAPDH as a substrate in arsenate buffer, as described previously.23,45 Briefly, we added 1 to 5 µg of cytosolic protein to 1 ml of assay buffer at 25°C and measured the rate of absorbance by continuous monitoring for 30 min at 340 nm using a spectrophotometer (Biomate 3; Thermo Scientific, Waltham, MA). The enzyme activity was expressed in nmol/min per mg of protein.

Immunoprecipitation
We immunoprecipitated total protein (500 µg) from tissue lysate with 4 µg of indicated antibody and 20 µl of protein A/G plus (Santa Cruz Biotechnology, Santa Cruz, CA) as per the manufacturer’s protocol. We resuspended the immunoprecipitated protein in SDS loading dye and performed Western blotting analysis as described previously.13,23,45 Using antibodies against PAR (BD Biosciences, San Jose, CA), GAPDH (Novus, Littleton, CO), or PARP (Trevena, Gaithersburg, MD), the signal was detected with ECL-plus system according to the manufacturer’s instructions (Amersham Biosciences, Piscataway, NJ). For biotinylated proteins, we performed blocking with 1% BSA in PBST, followed by biotin detection with 1:50,000 dilutions of streptavidin–horseradish peroxidase.

Immunofluorescence Microscopy
We performed immunofluorescence staining of LLCPK1 cells with primary antibodies against PAR (Trevena), AQP1 (Alpha Diagnostic, San Antonio, TX), or AQP7 (Santa Cruz Biotechnology) as described previously.39 Samples were viewed using a Leica DMR fluorescence microscope and the images were captured with an Optronics digital camera.

In Vitro Biotinylation Assay to Detect PARP Activity
We performed in vitro biotinylation to detect PARP activity in hypoxia-injured LLCPK1 cells as described previously.23,46 Briefly, 50 µg of protein was incubated with 60 µM biotinylated NAD in a 50-µl final volume of PARP reaction buffer (50 mM Tris-HCl [pH 8.0] and 25 mM MgCl2) at 37°C for 1 h. We terminated the reaction with SDS loading dye and subjected the samples to Western blot analysis to detect biotin expression, an indirect method to detect PAR expression.

ATP Assay
We performed ATP assay on tissue/cellular extracts using the Enlighten ATP assay system (Promega, Madison, WI) as previously described.13 Cellular ATP levels were expressed as nmol/µg protein.

Lactate Assay
We used the same tissue extracts used to measure ATP levels to measure lactate levels. We measured the tissue lactate levels using Lactate Assay Kit, EnzyChrom (BioAssay Systems, Hayward, CA) according
to the manufacturer’s protocol. The lactate levels were expressed in μM/μg protein.

We performed in vitro cell-free GAPDH assay as described previously, using unlabeled NAD⁺ or 6-biotin-17-NAD⁺ (biotin-NAD⁺), purified rabbit muscle GAPDH enzyme, activated DNA, and recombinant human PARP-1 enzyme (Trevigen, MD). The final reaction was performed in a reaction buffer (50 mM Tris-HCl [pH 8.0] and 25 mM MgCl₂) containing 5 μg of GAPDH, 5 μg of activated DNA, 10 μM biotin-NAD⁺ or 1 mM of NAD⁺, 20 or 200 μM GPI-15427, and 1 μl of PARP-1 enzyme (20 U). The sample was mixed well and incubated at 37°C for 1 h. Immediately after the reaction, we used 1 μl of the sample to measure GAPDH activity and mixed the rest of the sample with SDS loading dye. We performed Western blot analysis to detect biotin-labeled proteins, PAR, GAPDH, or PARP-1 with the indicated antibodies.

Cell Death Determination by LDH Release Assay

We performed LDH release assay using the cytotoxicity detection kit (Roche, Indianapolis, IN). We performed cell death determination by Trypan blue absorbance assay as described previously, with slight modifications. Briefly, we added 0.05% Trypan blue to each culture well at the end of each treatment and placed the plate in the incubator at 37°C for 15 min. We removed dye-containing medium by three washes with ice-cold PBS and lysed the cells with 1 ml of 1% SDS followed by absorbance measurements at 590 nm. The absorbance value from 2% Triton-X 100–treated cells was considered as a high value.

We used one-way ANOVA to compare the means of two different groups. P < 0.05 was considered statistically significant.

Statistical Analysis

All data are expressed as means ± SEM. We used unpaired t test to compare the means of two different groups. P < 0.05 was considered statistically significant.

REFERENCES


ACKNOWLEDGMENTS

This work was supported by a grant-in-aid from the American Heart Association to B.J.P., a research grant from Nebraska Kidney Association, and a graduate assistantship/fellowship from the University of Nebraska Medical Center to K.D.N.

We thank Dr. Pamela K. Carmines for critical reading of the manuscript and Dr. Jie Zhang, MGI Pharmaceuticals, for kindly providing the PARP inhibitor GPI-15427.

DISCLOSURES

None.
35. Gstraunthal G, Seppi T, Pfaller W: Impact of culture conditions, culture media volumes, and glucose content on metabolic properties of renal epithelial cell cultures: Are renal cells in tissue culture hypoxic?

Cell Physiol Biochem 9: 150–172, 1999

Supplemental information for this article is available online at http://www.jasn.org/.