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Research Article

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Transforming Growth Factor α Protection against Drug-induced Injury to the Rat Gastric Mucosa In Vivo

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Abstract

This study was designed to determine whether transforming growth factor α (TGF α) protects rat gastric mucosa against ethanol- and aspirin-induced injury. Systemic administration of TGF α dose-dependently decreased 100% ethanol-induced gastric mucosal injury; a dose of 50 $\mu\text{g}/\text{kg}$ delivered intraperitoneally 15 min before ethanol decreased macroscopic mucosal injury by > 90%. At the microscopic level, TGF α prevented deep gastric necrotic lesions and reduced disruption of surface epithelium. Pretreatment with orogastric TGF α (200 $\mu\text{g}/\text{kg}$) only partially (40%) decreased macroscopic ethanol damage. Intraperitoneal administration of TGF α at a dose of 10 $\mu\text{g}/\text{kg}$, which does not significantly inhibit gastric acid secretion, decreased aspirin-induced macroscopic damage by > 80%. TGF α protection does not seem to be mediated by prostaglandin, glutathione, or ornithine decarboxylase-related events, as evidenced by lack of influence of the inhibition of their production. Pretreatment with the sulfhydryl blocking agent *N*-ethylmaleimide partially abolished (40%) the protective effect of TGF α . In addition, systemic administration of TGF α resulted in a two-fold increase in tyrosine phosphorylation of phospholipase C-gamma 1 and in a time- and dose-dependent increase in levels of immunoreactive insoluble gastric mucin; these events occurred in a time frame consistent with their participation in the protective effect of TGF α . (*J. Clin. Invest.* 1992. 90:2409–2421.) Key words: ethanol • aspirin • prostaglandin • sulfhydryl • transforming growth factor α

Introduction

Epidermal growth factor (EGF)¹ protects gastric mucosa against a variety of ulcerogens (1–4). Transforming growth

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1. *Abbreviations used in this paper:* ASA, acetylsalicylic acid; DFMO, difluoromethylornithine; EGF, epidermal growth factor; EtOH, ethanol; INDO, indomethacin; NEM, *N*-ethylmaleimide; ODC, ornithine decarboxylase; PLC, phospholipase C; TGF α , transforming growth factor α ; TGF α /EGFr, TGF α /EGF receptor.

factor alpha (TGF α) is a polypeptide that shares with EGF structural homology (35%) (5, 6), a common membrane receptor, i.e., TGF α /EGF receptor (TGF α /EGFr) (7–9), and a qualitatively similar spectrum of biological activities (10). TGF α mRNA, but not EGF mRNA, has been shown to be expressed in the normal gastric mucosa from several species, including man (11). Also, TGF α mRNA and protein have been demonstrated to increase following acute injury to the rat stomach (12). Whether TGF α is protective to the gastric mucosa has not been studied. The present series of experiments was designed to determine whether systemic or orogastric administration of recombinant human TGF α afforded the rat gastric mucosa protection against an acid-independent form of acute injury (100% ethanol, EtOH) or an acid-dependent form of injury (aspirin, ASA). In the latter case, TGF α was administered intraperitoneally at a dose that was determined not to reduce gastric acidity significantly. In addition, we explored the mechanisms by which such protection may be conferred.

Methods

Male Sprague-Dawley rats (Harlan Sprague Dawley, Inc., Indianapolis, IN) weighing 180–220 g were used. Rats were housed individually in wire mesh cages to avoid coprophagy and fasted 24 h with ad lib. access to drinking water before the experiments.

Induction of gastric mucosal damage

EtOH-induced gastric mucosal damage was accomplished by the orogastric administration of 1 ml of 100% EtOH. Rats were killed 1 h later by cervical dislocation. Aspirin-induced gastric mucosal damage was accomplished by the orogastric administration of acetylsalicylic acid (Sigma Chemical Co., St. Louis, MO) (200 mg/kg body wt) suspended in 1 ml of 0.15 N HCl with the addition of two drops of Tween 80 (Sigma Chemical Co.) per 10 ml to keep ASA in a homogenous suspension. Rats were killed by cervical dislocation 4 h after ASA administration.

Assessment of damage

Gross. Excised stomachs were opened along the greater curvature and rinsed in saline. The degree of gastric mucosal damage was evaluated by using a computerized image analysis system (IM4–152; Analytical Imaging Concepts, Inc., Irvine, CA). The damage surface area was expressed as the percentage of the total glandular area.

Microscopic. The mucosal surface of each stomach was examined for the presence of hemorrhagic and necrotic lesions. Six tissue blocks were taken from each stomach from the areas of normal-appearing gastric mucosa. Specimens were fixed in buffered formalin and stained with hematoxylin and eosin plus periodic acid-Schiff. Coded mucosal specimens were evaluated qualitatively under light microscopy by an investigator (M. Romano) unaware of the treatment. In addition, the extent of deep histological necrosis (defined as necrotic lesions penetrating into the mucosa deeper than 0.2 mm) was quantitated morphometrically in a blinded fashion with the aid of planar morphometry computer software (Southern Micro Instruments, Inc., Atlanta, GA),

by measuring the length of mucosal strips and the length of necrotic lesions for each strip. Similarly, the disruption of continuity of the surface epithelium was quantitated by measuring the length of the mucosal strip and the length of mucosa devoid of the superficial epithelial layer. Results are expressed as a percentage of total mucosal strip length for each studied strip.

Scanning electron microscopy

Gastric mucosal specimens were fixed in 4% glutaraldehyde in 0.1 M phosphate buffer at pH 7.2. After a fixation period of 18 h, samples were dehydrated in a graded series of EtOH solutions and critically point dried by liquid CO₂ substitution. Samples were mounted on aluminum stubs using silver paste and coated with palladium gold. Specimens were then examined and photographed on a Hitachi 500 scanning electron microscope at a voltage of 20 kV.

Prostaglandin E₂ assay

Gastric juice. After a 24-h fast, rats were anesthetized with pentobarbital (50 mg/kg body wt intraperitoneally [i.p.]), the abdomen was opened, and a polyethylene catheter was introduced into the stomach through a small duodenal incision. The esophagus and the pylorus were ligated (the latter over the catheter). The stomach was washed three times with 2 ml of normal saline, and 1.5 ml of normal saline then was instilled. TGF α (200 μ g/kg) or normal saline (control) was administered i.p. and samples of the gastric contents (0.3–0.5 ml) were obtained 15, 30, and 60 min later. Samples were buffered with PBS pH 7.4, and frozen at -70°C until assayed. Later, samples were thawed, [³H]₂PGE₂ internal standard added, and PGE₂ quantified after extraction and purification using negative ion chemical ionization gas chromatography/mass spectrometry (13). Results are expressed as picograms per milliliter.

Gastric mucosa. The ex vivo generation of PGE₂ was determined according to described methods (14, 15). The animals were killed by cervical dislocation 30 min after i.p. administration of normal saline (control) or TGF α (200 μ g/kg). The stomachs were dissected out, opened along the greater curvature, rinsed in saline, and laid flat on absorbent paper to blot excess water. A portion of the corpus mucosa was peeled off, placed in a microfuge tube, weighed (\sim 150 mg), and chopped with fine scissors for 1 min in 1 ml of 0.1 M phosphate buffer, pH 7.4. The mixture was centrifuged (Centrifuge 5415 C; Eppendorf Inc., Fremont, CA) at 14,000 rpm for 30 s, the supernatant was discarded, and 0.5 ml of buffer was added. The pellet was dislodged and the tube vortexed for 1 min at room temperature. Indomethacin (INDO) (Sigma Chemical Co.) 50 μ g in 25 μ l of 1% NaHCO₃, was added to stop arachidonate metabolite generation. The mixture was centrifuged at 14,000 rpm for 30 s and the supernatant frozen for PGE₂ determination as described above. Results are expressed as nanograms per gram tissue.

Glutathione assay

Total GSH concentration was measured with the glutathione reductase-5,5'-dithiobis-(2-nitrobenzoic acid) recirculating assay of Tietze (16). In brief, animals were killed by cervical dislocation. The stomach was opened along the greater curvature and the gastric mucosa was scraped and weighed (\sim 30 mg). Tissue samples were homogenized in 10% TCA with a homogenizer (Polytron; Brinkmann Instruments, Inc., Westbury, NY). The homogenates were centrifuged at 1,500 g for 15 min at 4°C and an aliquot of the supernatant was neutralized with 0.3 M NaH₂PO₄. Neutralized samples were diluted (1:10) with 0.125 M Na₃PO₄, 6.3 mM EDTA, pH 7.5 (stock buffer); thereafter, 150 μ l of 0.3 mM NADPH (Boehringer Mannheim Biochemicals, Indianapolis, IN), 100 μ l of 6 mM dithiobis-(2-nitrobenzoic acid) (Sigma Chemical Co.), an aliquot of the sample, and stock buffer to give a final vol of 1 ml were added to cuvettes and the reaction was started by adding 10 μ l of glutathione reductase (\sim 50 U/ml) (Sigma Chemical Co.). The absorbance was monitored at 412 nm. The glutathione content of the aliquot assayed was determined by comparison with known amounts

of glutathione (Sigma Chemical Co.). Results are expressed as nanomoles per gram tissue.

Gastric secretion

Under ether anesthesia, the pylorus was ligated and the abdomen was closed. TGF α (1–100 μ g/kg) or normal saline was then injected i.p. 1 h after treatment, the animals were killed by cervical dislocation, the esophagus was ligated, and the stomach dissected out. Gastric juice was collected in graduated test tubes, its volume measured to the nearest 0.1 ml, and acid concentration determined by titration with 0.01 N NaOH to pH 7. The values are expressed in milliequivalents per liter (meq/liter) (acid concentration) and microequivalents per hour (μ eq/h) (acid output).

Determination of tyrosine phosphorylation of phospholipase C-gamma 1 (PLC- γ 1)

The method has been described previously (17). Scraped gastric mucosa was ground in a Polytron homogenizer (Brinkman Instruments, Inc.) on ice in hypotonic buffer (20 mM Hepes, pH 7.4, 5 mM EGTA, 1 mM MgCl₂, 1 mM PMSF, and 1 μ g/ml aprotinin, pepstatin, and leupeptin), centrifuged at 1,000 g for 10 min at 4°C, and the pellet discarded. A membrane fraction was obtained by centrifuging the supernatant at 100,000 g for 30 min at 4°C. The particulate membrane fraction was solubilized in a buffer containing 1% Triton X-100, 50 mM Hepes, pH 7.5, 50 mM NaCl, 50 mM NaF, 5 mM EDTA, 1 mM Na orthovanadate, 1 mM PMSF, and 1 μ g/ml aprotinin, pepstatin, and leupeptin. The supernatant cytosolic fractions from the ultracentrifugation were concentrated by lyophilization, reconstituted in the same buffer, and stored at -80°C until further use. For Western blots, membrane and cytosolic fractions (170 μ g) from gastric mucosa were subjected to 7% SDS-PAGE, transferred to nitrocellulose, and then incubated with a 1:500 dilution of the PLC- γ 1 antiserum followed by ¹²⁵I-donkey anti-rabbit IgG (\sim 200,000 cpm/ml; Amersham Corp., Arlington Heights, IL). The immunodetected PLC- γ 1 bands were visualized by autoradiography. For phosphotyrosine immunoprecipitation, a known amount of cytosolic protein (2 mg) from tissue extracts in a buffer containing phosphatase and protease inhibitors (see above), was absorbed onto 200 μ l of a Sepharose-linked antiphosphotyrosine (monoclonal 1G2) bead matrix (18) for 2–4 h at 4°C with rocking as described (19). After washing, the specifically absorbed phosphotyrosine proteins were eluted with 20 mM phenylphosphate, electrophoresed, and subjected to PLC- γ 1 immunoblot. To assess the specificity of the antiphosphotyrosine matrix, the tissue cytosols were immunoprecipitated with the antiphosphotyrosine matrix in the presence of excess phenylphosphate or phosphotyrosine before elution and PLC- γ 1 immunoblot. Protein was determined using the method described by Bradford (20).

Determination of ornithine decarboxylase (ODC) activity

Rat gastric mucosa was scraped away from the underlying smooth muscle with a glass slide. The tissues so obtained were then assayed for ODC activity and total protein. ODC assay was as described by Pegg and McGill (21). Tissues were placed in homogenization buffer (50 mM Tris-HCl, 100 μ M EDTA, 2.5 mM DTT, and 50 μ M pyridoxal phosphate), homogenized, sonicated, and centrifuged at 30,000 g at 4°C for 20 min. A 400- μ l aliquot of the supernatant was then incubated for 60 min at 37°C with homogenization buffer supplemented with 0.2 mM L-ornithine and 0.2 mM pyridoxal phosphate and with L-[¹⁴C]ornithine (0.25 μ Ci). The liberated ¹⁴CO₂ from the decarboxylation of ornithine was trapped on a piece of filter paper impregnated with 100 μ l of hyamine hydroxide, which was suspended in a center well above the reaction mixture. The reaction was stopped by the addition of 0.1 ml of 30% TCA; after an additional 30–60 min to collect residual radiolabeled CO₂, the filters were added to scintillation fluid and counted in a liquid scintillation counter. Aliquots of the 30,000 g supernatant were assayed for total protein, using the method described by Bradford (20). Results are expressed as picomoles ¹⁴CO₂ released per hour per milligram protein (pmol/h per mg).

Determination of gastric insoluble mucin

Adherent (insoluble) gastric mucin was gently removed from the gastric mucosa using a glass slide. PBS (1 ml) then was added and samples were stored frozen (-70°C) until assayed. Insoluble mucin was determined by an ELISA as described previously (22). In brief, 100 μl of mucin-containing sample was mixed with 100 μl of gastric mucin antibody in an Eppendorf tube (Brinkmann Instruments, Inc., Westbury, NY) and allowed to bind to equilibrium overnight at 37°C . At the same time, purified gastric mucin was added to each well of a 96-well plate (500 ng/well) and allowed to bind overnight. The next day the 96-well plate was rinsed with PBS three times and 300 μl of 0.5% crystalline grade BSA was added to each well and incubated for 1 h at 37°C . Each well was then washed with a "wash buffer" (PBS containing 0.05% Tween 20) three times and 75 μl of mucin-antibody mixture was added to the 96-well plate and incubated for 1 h at 37°C . Each well was rinsed again with wash buffer (three times) and 100 μl of biotinylated goat-anti-rabbit antibody (at a dilution of 1:2,000 in PBS containing 0.05% Tween 20) was added to each well and incubated for 1 h. Each well was rinsed again three times and incubated with 200 μl of streptavidin-horseradish peroxidase at a concentration of 625 ng/ml in PBS-Tween, pH 6.5 for 1 h. Each well was rinsed five times with wash buffer and color was developed using 2,2'-azino-bis (3-ethylbenzthiazoline-6-sulfonic acid) (0.01 g/25 ml) in 100 mM sodium citrate buffer, pH 4.2 plus 75 μl of 30% H_2O_2 /25 ml buffer (added immediately before use) for 5 min in the dark. The antibody binding was determined by reaching the color developed at 405 nm with a Titertek multiscan plate reader (Flow Laboratories, McLean, VA). Insoluble gastric mucin was expressed as nanograms per milliliter.

Experimental design

The experimental design is described in detail in the figure legends.

Statistical analysis

Results are expressed as mean \pm SEM. Significance of differences was assessed by Student's *t* test or analysis of variance followed by Duncan multiple range test (23) as appropriate. Differences were considered statistically significant if *P* was < 0.05 . Data which were expressed as percentage of control were analyzed before being normalized versus control.

Results

Effect of TGF α on EtOH- or ASA-induced damage. Orogastric administration of 100% EtOH is a well-characterized animal model that yields a reproducible degree of gastric mucosal injury (24, 25). This experimental model was selected because induction of mucosal injury is independent of luminal acidity (24, 26). As such, this model is able to test the gastric mucosal protective ability of the drug studied. Pretreatment with TGF α (200 $\mu\text{g}/\text{kg}$ intraperitoneally [i.p.]) provided nearly complete protection against gross mucosal injury to the rat glandular stomach (Fig. 1). TGF α administered i.p. at doses between 25 and 200 $\mu\text{g}/\text{kg}$ dose-dependently decreased gastric mucosal injury induced by EtOH ($r = -0.88$, $P < 0.05$) (Fig. 2), as quantitated by computerized image analysis. TGF α (200 $\mu\text{g}/\text{kg}$) decreased EtOH injury by more than 90% either when administered as a single pretreatment or when administered in four equally divided injections (Fig. 2). Pretreatment with TGF α (25 $\mu\text{g}/\text{kg}$ or 50 $\mu\text{g}/\text{kg}$) exerted a higher protective effect than that achieved by the same total dose given in repeated injections before and after EtOH (78 and 91% protection vs 27 and 63% protection, respectively (Fig. 2).

Unlike EtOH injury, ASA-induced gastric mucosal damage is acid dependent (27), i.e., the higher the acidity, the greater the damage. To standardize the effect of acid inhibition on

TGF α protection, we determined a dose of TGF α that did not significantly affect gastric secretion. Also, ASA was suspended in HCl 0.15 N (pH = 1.02). Fig. 3 shows a dose-dependent inhibition of gastric acid secretion by i.p. TGF α ($r = -0.86$, $P < 0.05$). At an i.p. dose of TGF α (10 $\mu\text{g}/\text{kg}$), which did not significantly affect gastric secretion (Fig. 3), there was 84.5% reduction in the damage induced by ASA (200 mg/kg-0.15 N HCl) ($P < 0.001$, Fig. 4). Repeated i.p. administration of TGF α 10 $\mu\text{g}/\text{kg}$ (30 min before and 1, 2, and 3 h after ASA) was not more effective than a single TGF α injection in preventing ASA injury (82.8% protection vs 84.5% protection, respectively, Fig. 4).

Kinetic and morphometric studies of TGF α 's protective effect. The EtOH-induced damage model was used in more detailed studies of the protective effect of TGF α . Fig. 5 shows the effect of orogastric administration of TGF α on EtOH-induced injury. TGF α (50 $\mu\text{g}/\text{kg}$) given 30 min before EtOH did not afford significant protection (Fig. 5). TGF α (200 $\mu\text{g}/\text{kg}$) decreased EtOH injury by only 40% ($P < 0.05$) (Fig. 5). Thus, in all subsequent studies, TGF α was administered i.p. The time-course of TGF α protection against EtOH injury is shown in Fig. 6. The protective effect was still significant (50% decrease in EtOH injury, $P < 0.01$) 6 h after the administration of TGF α (200 $\mu\text{g}/\text{kg}$) (Fig. 6).

Fig. 7, A-C, are representative photomicrographs that display the protective effect of TGF α on EtOH-induced injury. The typical histological appearance of normal gastric mucosa is shown in Fig. 7 A. EtOH induced complete disruption of the superficial epithelium and caused necrosis in the upper region of the gastric glands (Fig. 7 B). In contrast, the gastric mucosa appeared to be well preserved if rats were pretreated with TGF α (200 $\mu\text{g}/\text{kg}$ i.p.) 30 min before EtOH challenge (Fig. 7 C). These sections were taken from areas that appeared grossly normal. The extent of disrupted surface epithelium and deep mucosal necrosis was quantified by planar morphometry in a blinded fashion and expressed as percentage of total mucosal strip length (Fig. 8; see Methods). Surface epithelial disruption was partially reduced by TGF α (22.4% \pm 0.7 vs 47.1% \pm 5.2 in controls, $P < 0.01$) (Fig. 8). In addition, pretreatment with TGF α significantly reduced EtOH-induced deep necrosis (2.9% \pm vs 13.2% \pm 1.9 in controls, $P < 0.01$).

Fig. 9 (A-C) shows the scanning electron microscopy of normal rat gastric mucosa (Fig. 9 A) and of EtOH-injured mucosa from saline-pretreated (Fig. 9 B) and TGF α -pretreated rats (Fig. 9 C). Once again, samples were taken from areas which appeared normal upon gross examination. In control animals, EtOH caused severe disruption of the surface epithelium that resulted in formation of areas of denuded lamina propria (Fig. 9 B). Pretreatment with TGF α (200 $\mu\text{g}/\text{kg}$) partially prevented EtOH injury to the superficial epithelium (Fig. 9 C).

Effect of systemic TGF α on tyrosine phosphorylation of PLC- γ 1. We considered whether the protection afforded by TGF α was mediated through activation of the TGF α /EGFr in the gastric mucosa. We studied the effect of systemic TGF α on tyrosine phosphorylation of PLC- γ 1, a putative substrate for the TGF α /EGFr (28, 29). Cytosolic and membrane fractions of scraped gastric mucosa were examined for PLC- γ 1 by Western blot analysis. The cytosolic fractions contained the majority of PLC- γ 1 (Fig. 10 A) (data related to membrane fractions are not shown). To detect the tyrosine phosphorylation species of PLC- γ 1, 2 mg of cytosolic protein was absorbed onto phos-



Figure 1. Macroscopic appearance of rat gastric mucosa after orogastric administration of 100% EtOH in normals saline (NS)- and TGF α -pretreated animals. TGF α (200 μ g/kg) or normal saline was administered i.p. 30 min before EtOH.

photyrosine-Ab matrix, eluted with phenylphosphate, and PLC- γ 1 examined by Western blot analysis. TGF α (200 μ g/kg) caused a time-dependent increase in tyrosine phosphorylation of PLC- γ 1 in the gastric mucosa (Fig. 10 B). At 15 min, there was a twofold increase in tyrosine phosphorylation of PLC- γ 1, as assessed by measurement of relative optical density (data not shown). These findings were confirmed in two additional experiments (data not shown). Thus systemic administration of TGF α results in a biochemical event in the gastric mucosa linked to postreceptor signaling that occurs in a time frame consistent with its participation in TGF α -mediated gastric protection.

Role of endogenous prostaglandin in TGF α protection. We next examined possible mechanisms by which TGF α might exert its protective effect. Prostaglandins are considered to play a role in the ability of the gastric mucosa to resist different forms of injury (24). TGF α (200 μ g/kg) did not increase PGE $_2$ release into the gastric juice nor did it increase gastric tissue levels of PGE $_2$ (Fig. 11 A), as assessed by mass spectrometry. Also, TGF α was not able to counteract INDO-induced depletion of PGE $_2$ in the gastric mucosa (Fig. 11 B). To further explore the role of endogenous prostaglandins in TGF α protection, we studied whether pretreatment with INDO (5 mg/kg), a concentration which decreases PGE $_2$ content of the rat gastric

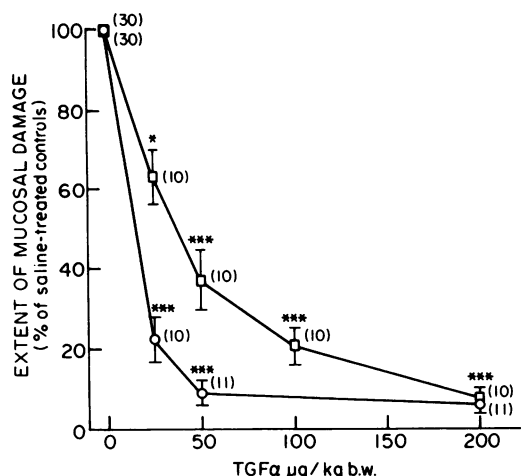


Figure 2. Effect of systemic administration of TGF α on EtOH-induced rat gastric mucosal injury as quantitatively assessed by computerized image analysis. Rats were pretreated with i.p. injection of TGF α (25–200 μ g/kg) or normal saline and, after 15 min, were given orogastric 100% EtOH (circles). In another series of experiments the total dose of TGF α was divided into four equal doses which were administered i.p. 15 min before and 15, 30, and 45 min after orogastric 100% EtOH (squares). Animals were killed 60 min after EtOH. The damaged area in the control groups is 22.9% \pm 2.1 of the total glandular area. Mean \pm SE of (n) rats per each study group. \square , Divided doses; \circ , single dose. * P < 0.05 vs control; *** P < 0.001 vs control.

mucosa by > 70% (Fig. 11 B), was able to prevent TGF α -induced protection. INDO caused a slight increase in EtOH-induced injury, but this did not reach statistical significance (Fig. 12). Pretreatment with INDO did not abolish the protective effect of TGF α (Fig. 12).

Role of ornithine decarboxylase activity in TGF α protection. Recently, an increase in ODC activity has been shown to be associated with the ability of EGF to protect rat gastric mucosa against stress-induced injury (30). TGF α (100 μ g/kg) administered i.p. increased ODC activity in the rat gastric mucosa from 14.7 \pm 2.7 to 22.0 \pm 4.0 pmol/h per mg. The ODC inhibitor difluoromethylornithine (DFMO), at a concentration that decreased basal ODC activity by 65% (P < 0.01) and counteracted TGF α -stimulated increase in ODC activity (51% inhibition, P < 0.01), did not prevent the protective effect of TGF α (Fig. 13). Thus TGF α -induced gastric mucosal protec-

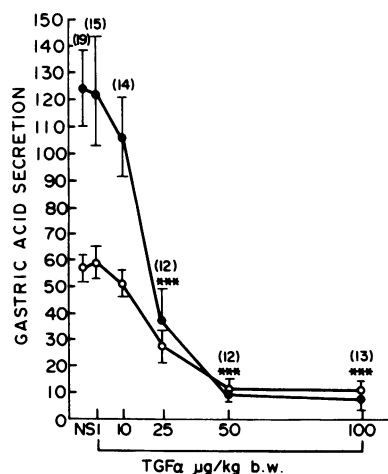


Figure 3. Effect of TGF α on gastric acid secretion. Gastric acid secretion was measured in pylorus-ligated rats 1 h after i.p. administration of TGF α (1–100 μ g/kg) or normal saline. Mean \pm SE of (n) rats per each study group. \bullet , Acid output (μ eq/h); \circ , acid concentration (meq/liter); *** P < 0.001 vs NS.

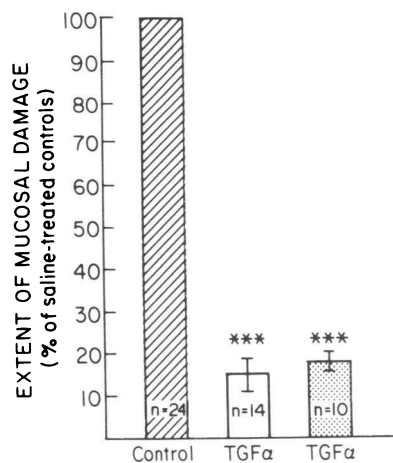


Figure 4. Effect of TGF α on acidified aspirin (ASA)-induced damage to the rat gastric mucosa. Gross mucosal injury was quantitated by computerized image analysis 4 h after ASA (200 mg/kg) in rats pretreated with i.p. injection of TGF α (10 μ g/kg) or normal saline 30 min before ASA. In another series of experiments, rats were treated with TGF α (10 μ g/kg) or normal saline 30 min before and 1, 2, and 3

h after ASA administration. Mean \pm SE of (n) rats per each study group. \square , TGF α 10 μ g/kg body wt; \square , TGF α 10 μ g/kg body wt \times 4; *** P < 0.001 vs control.

tion appears to be mediated by nonprostaglandin and non-ODC related signaling pathways.

Role of endogenous sulfhydryls in TGF α protection. Sulfhydryl compounds also have been demonstrated to be involved in the protection of the gastric mucosa against various forms of injury (31–33). TGF α did not stimulate glutathione synthesis in the stomach (Fig. 14 A). To evaluate further the effect of TGF α on glutathione metabolism, we evaluated whether TGF α prevented the depletion of gastric glutathione induced by phorone, an electrophilic agent which acts through glutathione-S-transferases (34). Phorone (50–250 mg/kg) decreased gastric glutathione tissue levels in a dose-dependent manner ($r = -0.97$, P < 0.05), (data not shown). Pretreatment with TGF α (200 μ g/kg) did not prevent the glutathione-depleting effect of phorone (125 mg/kg) (Fig. 14 B).

To assess further the role of endogenous sulfhydryl compounds in TGF α -mediated protection, we studied whether the sulfhydryl group alkylator *N*-ethylmaleimide (NEM) counteracted TGF α 's protective effect. Fig. 15 shows that NEM (10 mg/kg), a concentration which did not significantly increase EtOH injury to the gastric mucosa, decreased the protective effect of TGF α by 40%. NEM (20 mg/kg) abrogated the protective effect of TGF α . However, NEM at this concentration

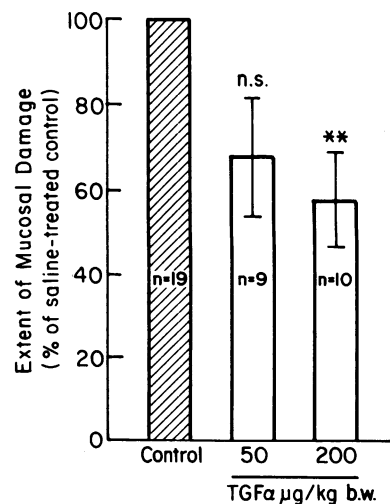


Figure 5. Effect of orogastric administration of TGF α on EtOH-induced injury. TGF α (50 and 200 μ g/kg) or normal saline was administered orogastrically 30 min before EtOH. The damaged area in the control group is 17.0% \pm 4.0 of the total glandular area. Mean \pm SE of (n) rats per each study group. n.s., not significant vs control; ** P < 0.01 vs control.

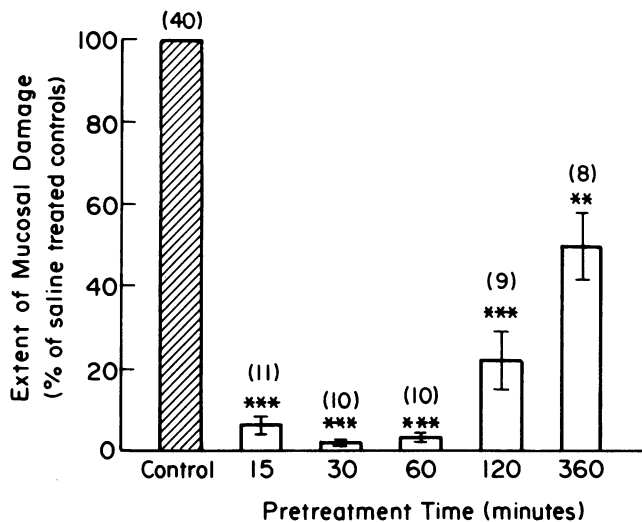


Figure 6. Time sequence of TGF α protection against EtOH injury. TGF α (200 μ g/kg) was administered i.p. 15 min to 6 h before EtOH administration. The damaged area in the control group is 25.9% \pm 4.0 of the total glandular area. Mean \pm SE of (n) rats per each study group. *** P < 0.001 vs control.

significantly affected EtOH-induced mucosal injury by increasing the damage to the glandular area from 20.4% \pm 2.1 to 34.7% \pm 6.4 (P < 0.05) (Fig. 15). NEM (10 and 20 mg/kg) did not affect glutathione tissue levels (data not shown).

Effect of TGF α on gastric insoluble (adherent) mucin. We also examined the effect of TGF α on levels of gastric mucin. In these studies, the gastric mucosa was scraped lightly with a glass slide at the indicated times, and mucin levels determined by a reverse ELISA with an antibody that recognizes biologically active, insoluble gastric mucin (22). The gastric mucosa was intact microscopically after light scraping of both TGF α and normal saline-treated rats (data not shown). TGF α (100 μ g/kg) time-dependently increased gastric insoluble mucin (Fig. 16 A). At 15 and 30 min from TGF α administration, there was a 7.3-fold (P < 0.001 vs control) and 14.5-fold (P < 0.001 vs control) increase in adherent mucus, respectively (Fig. 16 A). TGF α (1–100 μ g/kg) increased gastric adherent mucin in a dose-dependent manner ($r = 0.987$, P < 0.05) (Fig. 16 B).

Discussion

EGF protects gastric mucosa against damage induced by ASA (1, 2), cysteamine (3), EtOH (4, 35–37), and stress (30). EGF is localized mainly in submandibular salivary glands and in Brunner's glands (38, 39). Induction of a novel EGF-secreting cell lineage has been shown adjacent to ulcerated human gastrointestinal mucosa (40); however, it is controversial whether EGF is expressed in the normal gastric mucosa (11, 38, 39, 41–43). TGF α is a 50-amino acid polypeptide (44) which shares with EGF structural homology (5, 6), a common receptor (7–9) and a nearly identical spectrum of biological activity (10). In particular, both EGF and TGF α stimulate proliferation of gastric epithelial cells in vitro (45) and inhibit gastric acid secretion in vitro (46–48) and in vivo (49, 50). Unlike EGF, TGF α has been demonstrated to be expressed in the normal gastric mucosa of different species, including man and rat (11, 43, 51). Also, immunoreactive TGF α is present in the normal human stomach in significantly higher levels than im-

munoreactive EGF (43). Recently, TGF α mRNA and protein have been shown to increase after acute gastric injury in the rat (12), thus suggesting a role for TGF α in gastric mucosal repair. Whether TGF α is able to prevent different forms of injury to the gastric mucosa has not been studied. Therefore, we evaluated the effect of TGF α on EtOH- or ASA-induced damage to the rat gastric mucosa.

Systemic administration of TGF α prevented EtOH injury in a dose-dependent manner and decreased EtOH-induced gross mucosal damage by > 90% (Fig. 2). A single pretreatment with systemic TGF α was as effective as repeated administration (Fig. 2). At the microscopic level, pretreatment with TGF α prevented by > 75% deep gastric mucosal necrosis and reduced by > 50% disruption of surface epithelium induced by EtOH (Fig. 8). We also evaluated whether orogastric TGF α exerted any protective effect against EtOH injury. TGF α (50 μ g/kg), a dose that affords a > 90% protection when administered systemically, did not significantly prevent EtOH injury. At an oral dose of 200 μ g/kg, TGF α provided only 40% protection to the rat gastric mucosa against EtOH-induced damage (Fig. 5). It remains controversial whether the TGF α /EGFr, which has been demonstrated to be located on the basolateral membrane of target cells, is also expressed on the cell surface (52–54). Systemic EGF inhibits gastric acid secretion in rats while orogastric EGF does not (55). Likewise, serosal, but not luminal EGF, inhibits acid secretion from guinea pig gastric mucosa mounted in Ussing chambers (46). Moreover, it is well known that acidic pH impairs the binding of TGF α to its receptor (8, 56). Therefore, we speculate that the partial protection obtained with orogastric administration of TGF α reflects a systemic effect secondary to the absorption of TGF α with subsequent delivery to TGF α /EGFr located on the basolateral membrane.

ASA-induced gastric mucosal damage is dependent on the pH of gastric contents (the more acid, the more damage) (27), whereas EtOH injury is not (24, 26). Therefore, to rule out the influence of gastric acid inhibition on protection, we used a concentration of TGF α (10 μ g/kg) that did not significantly affect gastric secretion (Fig. 3). Moreover, ASA was suspended in 0.15 N HCl (pH = 1.02). TGF α (10 μ g/kg) decreased acidified ASA-induced damage by > 80% (Fig. 4). Repeated administration of TGF α (10 μ g/kg) (30 min before and 1, 2, and 3 h after ASA) did not show any significant improvement in gastric mucosal protection as compared to a single injection of TGF α (10 μ g/kg) 30 min before ASA administration. Therefore, TGF α is able to protect the gastric mucosa from a necrotizing agent such as EtOH whose damaging effect is acid independent and, in a non-antisecretory dose, from ASA-induced injury that is acid-dependent. Thus, TGF α may be considered a true gastric mucosal protective agent.

We did not directly compare the gastric mucosal protective ability of TGF α and EGF. However, previous studies have shown that mouse EGF (100 μ g/kg) given orogastrically or parenterally only partly reduces the mucosal damage caused by absolute EtOH (35, 37). Also, recombinant human EGF (30 μ g/kg) has recently been shown not to exert any protective effect when given 6 h before 50% EtOH (4). On the contrary, in our study, a 50% protective effect against EtOH injury was still seen 6 h after i.p. administration of TGF α (Fig. 6). These previous studies, if corroborated, suggest that TGF α , even though acting through the same receptor as EGF, seems more efficient than EGF in gastric mucosal protection. This is not altogether

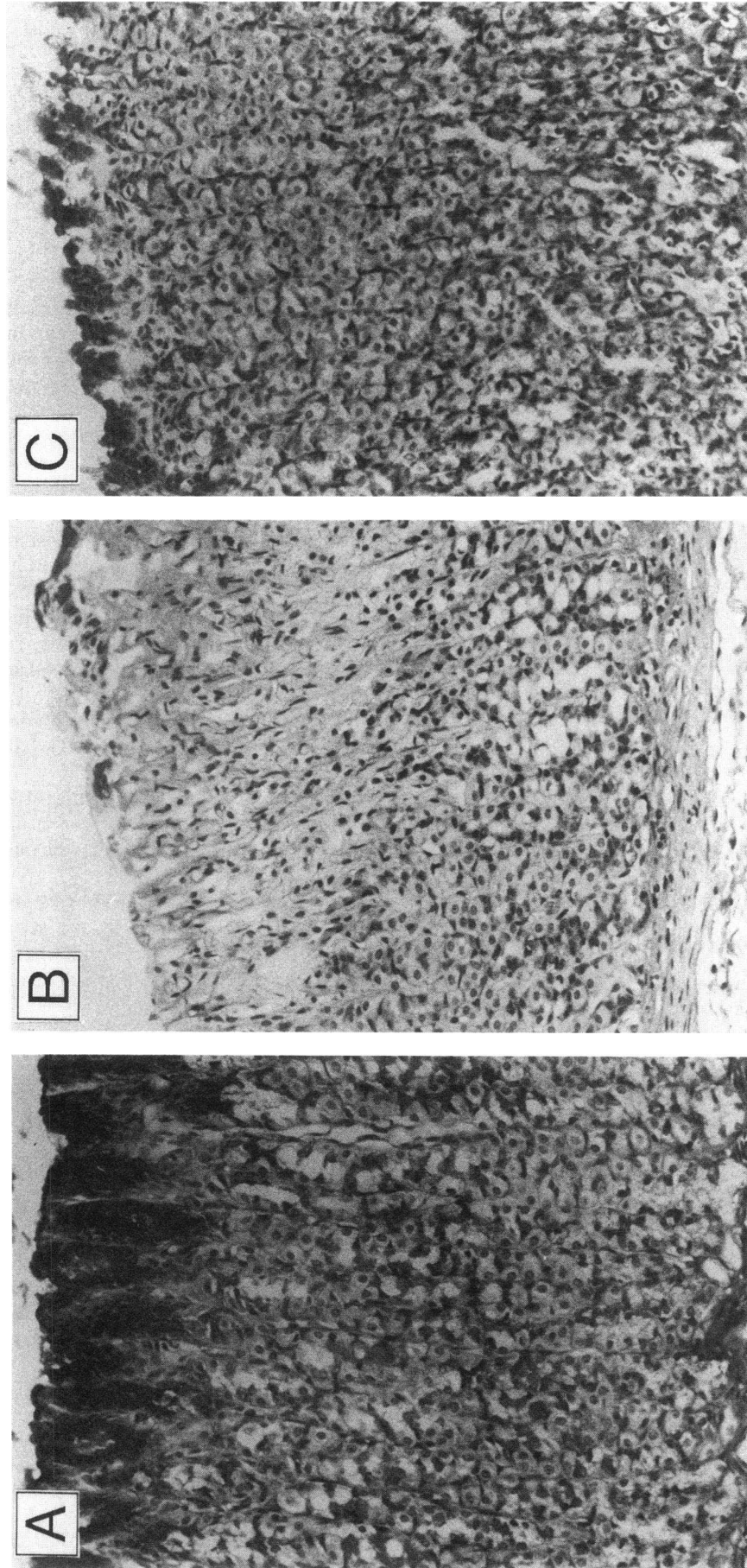


Figure 7. (A-C) Histological effect of $TGF\alpha$ on EtOH injury. A represents normal rat gastric mucosa. EtOH induced disruption of the superficial epithelium and caused deep necrotic damage (B). Pretreatment with $TGF\alpha$ ($200 \mu\text{g}/\text{kg}$) partially preserved surface cells from EtOH injury and prevented deep glandular necrosis (C). Specimens were taken from areas which appeared normal upon gross examination. Sections were stained with hematoxylin and eosin and periodic acid-Schiff. $\times 66$.

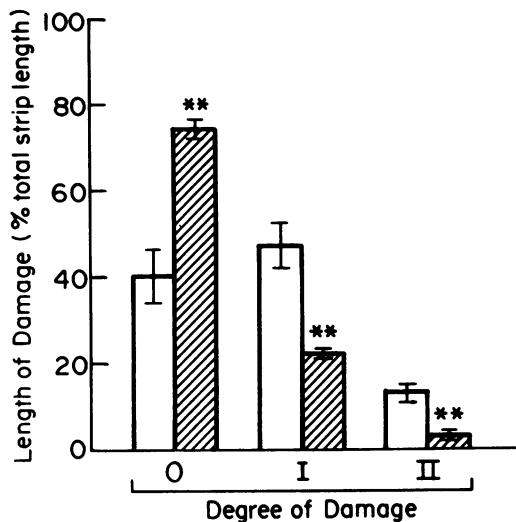


Figure 8. Quantitation of deep gastric mucosal necrosis and disruption of gastric surface epithelium in TGF α -pretreated and control rats after EtOH administration. Rats were given 100% EtOH 30 min after i.p. administration of TGF α (200 μ g/kg) or normal saline (control). Results are expressed as percentage of total mucosal strip length and represent the mean \pm SE of six rats per each study group. 0, no damage; I, disruption of surface epithelium; II, deep necrosis (> 0.2 mm). Specimens were from macroscopically noninjured gastric mucosa. Total mucosal strip length was 9.5 \pm 0.4 mm in controls and 9.7 \pm 0.8 mm in TGF α -pretreated animals. \square , Normal saline; \blacksquare , TGF α 200 μ g/kg body wt; ** P < 0.01 vs normal saline.

surprising, since several quantitative differences have been observed in the biological activity of TGF α and EGF (10). For example, TGF α has been shown to be more potent than EGF in inducing angiogenesis in hamster cheek pouches (58). Also, TGF α is more effective than EGF in increasing regional blood flow in the anesthetized dog (59) and in stimulating osteoclast precursor cells (60). These quantitative differences may, in part, be explained by differential processing of the ligand-receptor complexes (61). Thus far, no distinct TGF α receptor has been identified. However, to examine the comparative protective effects of TGF α and EGF more rigorously, we are presently comparing the effect of recombinant human forms of TGF α and EGF on EtOH- and ASA-induced damage to the rat gastric mucosa.

Levels of endogenous rat TGF α protein have been measured by a sensitive and specific rat RIA in the gastric mucosa and juice as well as in the plasma (14.4 \pm 4.8 pg/ μ g DNA, 0.3 \pm 0.1 total ng, 88 pg/ml, respectively) (Dempsey, P. J., and R. J. Coffey, unpublished observations). 15 min after i.p. administration of TGF α (50 μ g/kg), plasma levels of human TGF α were 12 \pm 2.0 ng/ml, as determined by a human TGF α RIA that does not recognize rat TGF α (Dempsey, P. J., and R. J. Coffey, unpublished observations). Therefore, the doses used in this study are pharmacological ones. This must, however, be tempered by the realization that the effective concentration of the endogenously produced TGF α at the cell surface might be far greater than that measured in the gastric tissues or plasma.

The biological effects of TGF α are mediated through binding to the TGF α /EGFr (62). Accumulating evidence indicates that PLC- γ 1 is a substrate for the tyrosine kinase of the TGF α /EGFr (28, 29). The present study demonstrates for the first

time in vivo in nonneoplastic tissues that systemic administration of TGF α results in a time-dependent increase in tyrosine phosphorylated PLC- γ 1 (Fig. 10). Additional biological relevance of this finding derives from the observation by Konda et al. in which prostaglandin protection of isolated guinea pig chief cells against EtOH has been shown to be via an increase in diacylglycerol (63), which is derived from the PLC- γ 1-induced breakdown of the phosphoinositide (64), thus implicating activation of PLC- γ 1 as a mediator of gastric protection.

Prostaglandins are known to play an important role in the protection of gastric mucosa (24). Therefore, we studied whether TGF α protection might be mediated by endogenous prostaglandins. TGF α , at a protective concentration, did not stimulate PGE $_2$ production by the rat gastric mucosa nor did it counteract the INDO-induced depletion in gastric PGE $_2$ (Fig. 11). Furthermore, TGF α protection was not prevented by pretreatment with INDO in a concentration which decreased gastric tissue levels by > 70% PGE $_2$ (Fig. 12). Therefore, TGF α , like EGF (1, 2), does not seem to protect gastric mucosa through stimulation of endogenous prostanoid synthesis. A PGE $_1$ analogue (i.e., misoprostol) recently has been demonstrated to be effective in preventing ASA-induced injury to the human gastric mucosa (65) and approved by the FDA specifically for use in the prevention of nonsteroidal antiinflammatory drug-induced gastric mucosal damage. The observation that TGF α , at a non-antisecretory dose, protects the gastric mucosa against ASA-induced damage and appears to act independently of prostaglandins, leads us to suggest that TGF α might prove of use, alone or in combination with a prostaglandin derivative, to attenuate gastric injury induced by nonsteroidal antiinflammatory drugs (66).

Recently, protection by EGF against stress-induced gastric lesions in the rat has been shown to be mediated in part by an increase in the activity of ODC (30), the rate-limiting enzyme in the biosynthesis of polyamines (67), which also play a role in gastric mucosal protection (68). Pretreatment with TGF α caused a 1.5-fold increase in ODC activity in the gastric mucosa. However, DFMO, a specific and irreversible inhibitor of ODC (69), at a concentration which significantly inhibited basal as well as TGF α -stimulated ODC activity, did not counteract the protective effect of TGF α (Fig. 13). Therefore, it is unlikely that the protective effect of TGF α is mediated through an ODC-related pathway.

Sulfhydryl compounds protect gastric mucosa against damage induced by different ulcerogens in vivo (31-33) and in vitro (70). In particular, glutathione, the most abundant thiol in cells (34), has been shown to play an important role in gastric mucosal protection (71-73), even though this has been questioned by other authors (74). Our study indicates that TGF α does not stimulate glutathione synthesis in the rat gastric mucosa nor does it prevent the depletion of glutathione gastric tissue levels induced by phorone (Fig. 14). However, pretreatment with the sulfhydryl group alkylator NEM, at a concentration which did not increase EtOH injury, partially (40%) prevented TGF α -induced protection (Fig. 15). NEM, at a concentration that significantly increased EtOH injury, almost completely (76%) abolished the protective effect of TGF α (Fig. 15). The inhibition of the protective effect of TGF α by NEM does not seem to be accounted for by interference with the binding of TGF α to its receptor. In fact, NEM up to 5 μ M did not interfere with TGF α binding to the TGF α /EGFr in A431 cells (Romano, M., and R. J. Coffey, unpub-

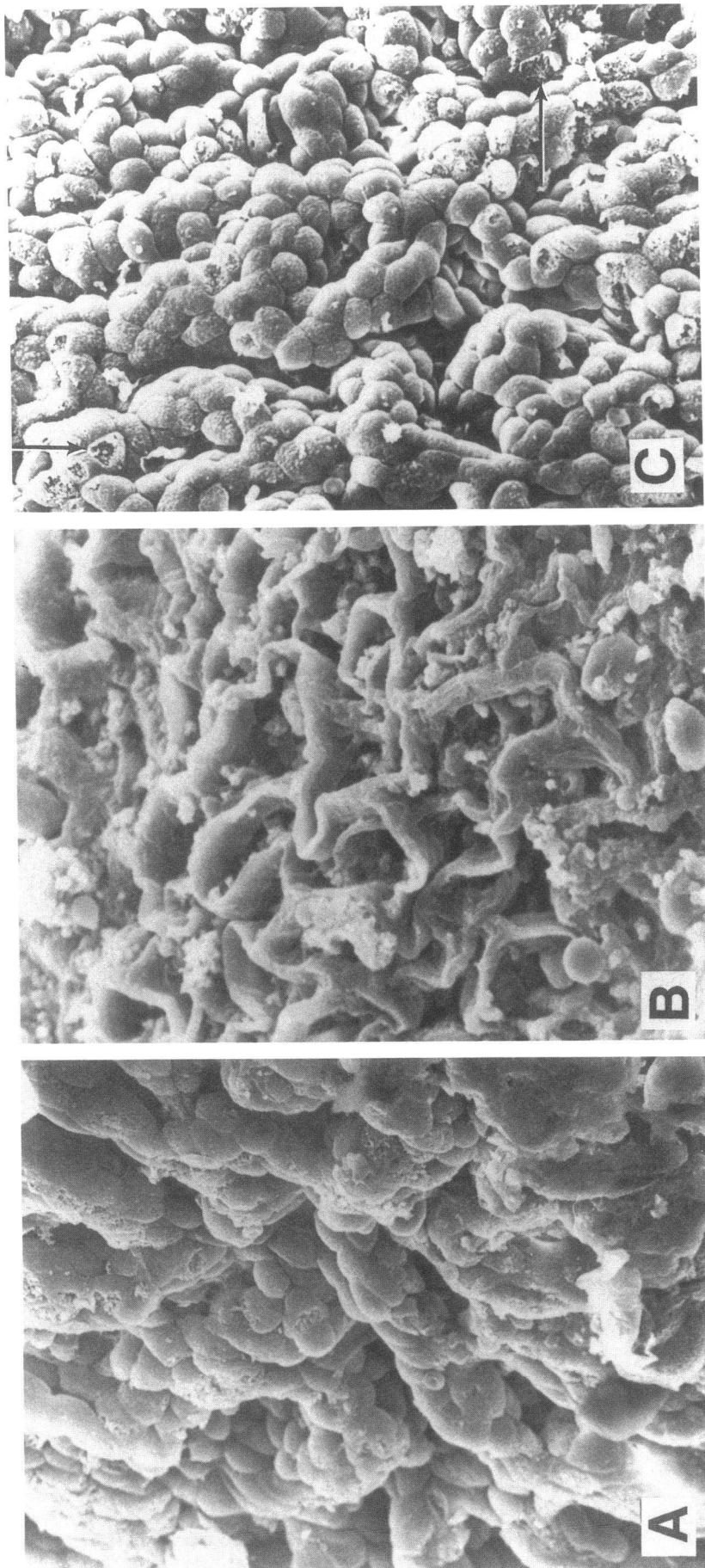


Figure 9. (A-C) Effect of TGF α on EtOH injury by scanning electron microscopy. A represents normal gastric mucosa. EtOH completely disrupted surface mucosal cells and caused denudation of lamina propria in normal saline-treated animals. (B) Pretreatment with systemic TGF α (200 μ g/kg) partially prevented damage to superficial epithelium. (C) The arrows indicate cells with some degree of damage. Specimens were taken from normally appearing areas located between hemorrhagic bands. $\times 800$.

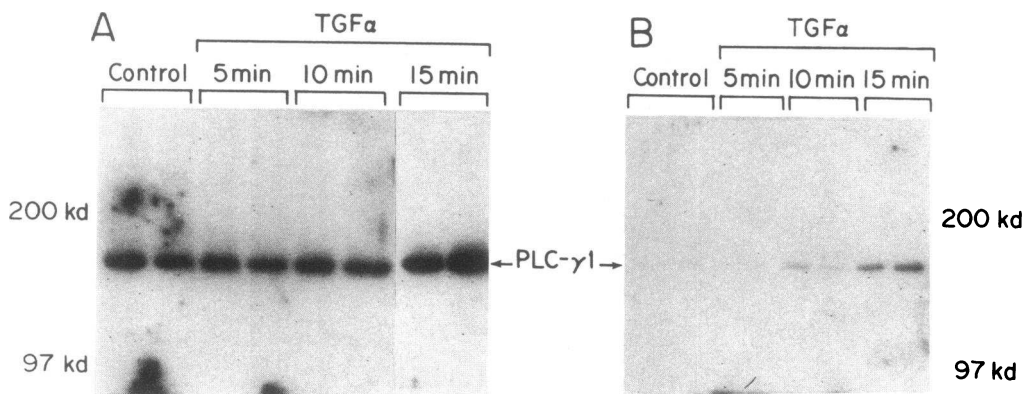


Figure 10. Effect of systemic TGF α on tyrosine phosphorylation of PLC- γ 1. Tyrosine phosphorylation of PLC- γ 1 was determined in the scraped gastric mucosa 5, 10, and 15 min after i.p. administration of TGF α (200 μ g/kg) or normal saline (control) by immunoprecipitation with an antiphosphotyrosine antibody followed by Western blot analysis using an antibody to PLC- γ 1. (A) Total cytoplasmic PLC- γ 1; (B) cytoplasmic PLC- γ 1 after immunoprecipitation of 2 mg of cytosol with an antiphosphotyrosine antibody.

lished observation). Moreover, NEM, at the concentrations that partially reversed TGF α 's protection, did not affect glutathione tissue levels. Therefore, TGF α 's protection may be mediated partly by nonprotein sulfhydryls other than glutathione

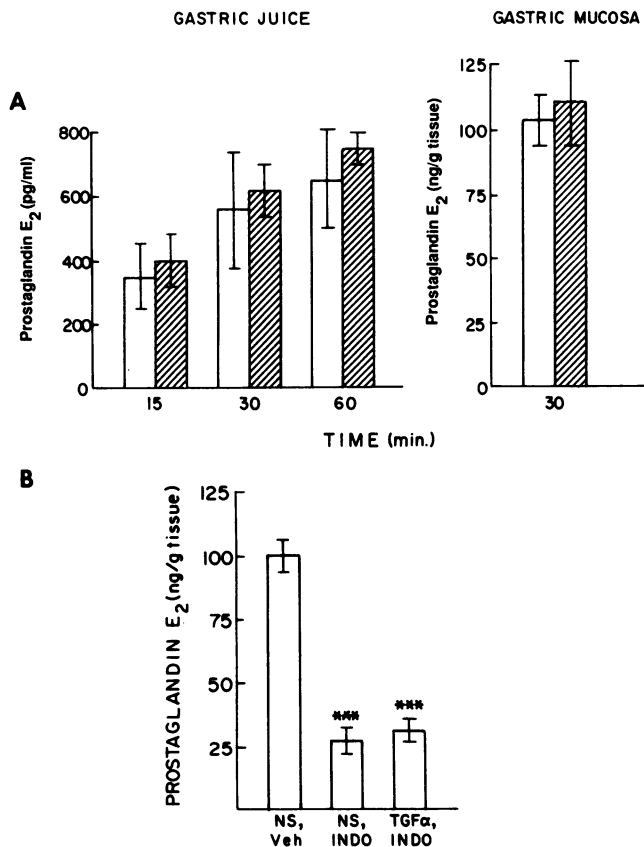


Figure 11. Effect of TGF α on prostaglandin E₂ metabolism in the rat gastric mucosa. (A) PGE₂ was measured in the gastric juice and in the gastric mucosa at different time intervals after i.p. administration of TGF α (200 μ g/kg) or normal saline. Mean \pm SE of four animals per each study group. \square , Control (normal saline); \blacksquare , TGF α 200 μ g/kg body wt. (B) 30 min after i.p. injection of TGF α (200 μ g/kg) or normal saline, rats were given INDO (5 mg/kg) or 5% NaHCO₃ (vehicle for INDO) subcutaneously and were killed 1 h later. PGE₂ was measured in the gastric mucosa. Mean \pm SE of (n) animals per each study group. ****P* < 0.001 vs NS, Veh.

or through a protein-bound, sulfhydryl-sensitive metabolic pathway (75). However, the possibility exists that the reversal of TGF α protection by NEM is caused by an increase in the microvascular permeability which renders the gastric mucosa more vulnerable to the damaging effect of EtOH (76).

In the attempt to elucidate further the mechanism of TGF α protection, we studied the effect of TGF α on insoluble (adherent) gastric mucin. TGF α dose- and time-dependently stimulated adherent gastric mucus (Fig. 16). At 15 min from TGF α administration, there was a sevenfold increase in insoluble mucin, which is consistent with the time frame of TGF α -induced mucosal protection. However, the role of gastric mucus as a protective barrier on the gastric mucosa is controversial (77–79). Adherent mucus is in fact permeable to damaging agents such as EtOH and ASA (80) which gain access through the gel to the superficial epithelial cells. On the other hand, removal of the gelatinous layer of mucus and cellular debris which formed after exposure of the gastric mucosa to 70% EtOH inhibited the protection against a rechallenge with the same necrotizing agent (81). We postulate that the TGF α -induced increase in the adherent mucus gel layer covering the epithelial surface

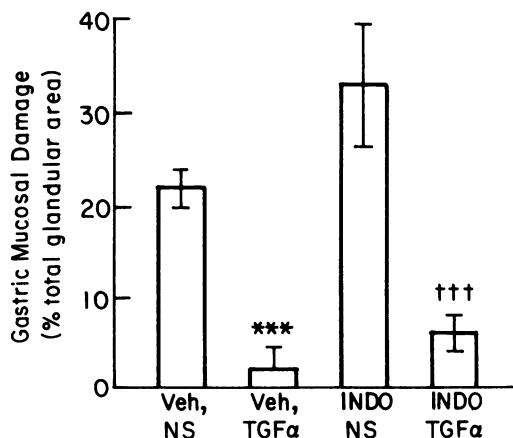


Figure 12. Effect of INDO on TGF α -induced protection against EtOH injury. Rats were pretreated with INDO (5 mg/kg) or 5% NaHCO₃ subcutaneously; after 1 h, rats were given TGF α (200 μ g/kg) or normal saline i.p. and, 30 min later, both groups received orogastric 100% EtOH. Mean \pm SE of eight rats per each study group. ****P* < 0.001 vs Veh, NS; †††*P* < 0.001 vs INDO, NS.

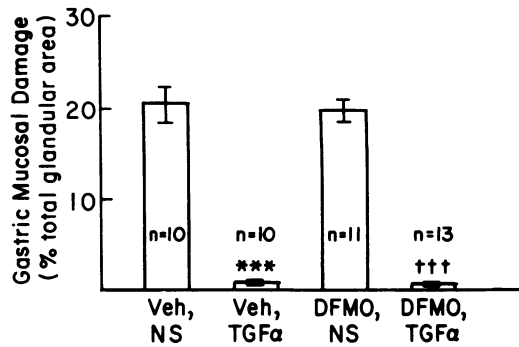


Figure 13. Effect of DFMO on TGF α -induced protection. Rats were pretreated with DFMO (400 mg/kg) or dH₂O (vehicle for DFMO) i.p.; after 4 h rats were given i.p. injection of TGF α (200 μ g/kg) or normal saline and, 30 min later, orogastric 100% EtOH. Mean \pm SE of (n) rats per each study group. ****P* < 0.001 vs Veh, NS; †††*P* < 0.001 vs DFMO, NS.

may act as a dilutional barrier to damaging agents, may delay and/or restrict further damage induced by acid and pepsin, and may accelerate early reparative events. An alternate mechanism by which mucin might protect the gastric mucosa is through its ability to scavenge toxic oxygen metabolites (82) which are generated by EtOH and ASA (83). The rapid increase in mucin levels is likely caused by release of preformed mucin. Studies are underway to examine the effect of TGF α on rat gastric mucin mRNA expression and protein production.

In conclusion: (a) systemic TGF α protects the rat gastric mucosa against EtOH-induced microscopic and macroscopic injury; (b) this protective effect is seen also with ASA-induced gastric injury at non-antisecretory concentrations of TGF α ; (c) this protection does not seem to be mediated by prostaglandin,

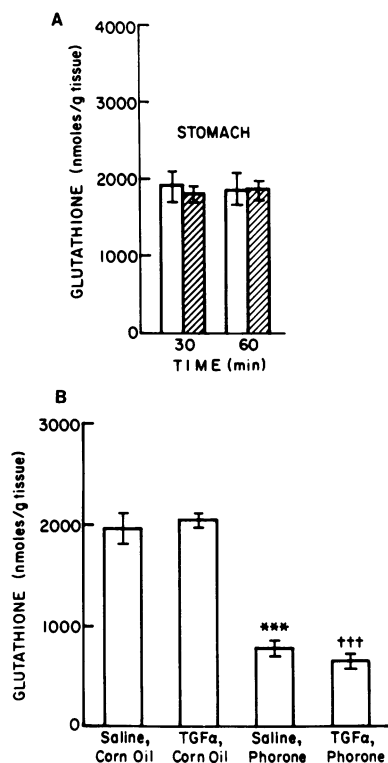


Figure 14. Effect of TGF α on gastric glutathione metabolism. (A) Glutathione was measured in the scraped gastric mucosa 30 and 60 min after i.p. administration of TGF α (200 μ g/kg) or normal saline. Mean \pm SE of four rats per each study group. □, Control (normal saline); ■, TGF α 200 μ g/kg body wt. (B) Rats were pretreated with i.p. injection of TGF α (200 μ g/kg) or normal saline; after 30 min rats were given phorone (125 mg/kg) or corn oil i.p. and were killed 1 h later. Glutathione was measured in the scraped gastric mucosa. Mean \pm SE of four rats per each study group. ****P* < 0.001 vs saline, corn oil; †††*P* < 0.001 vs TGF α , corn oil.

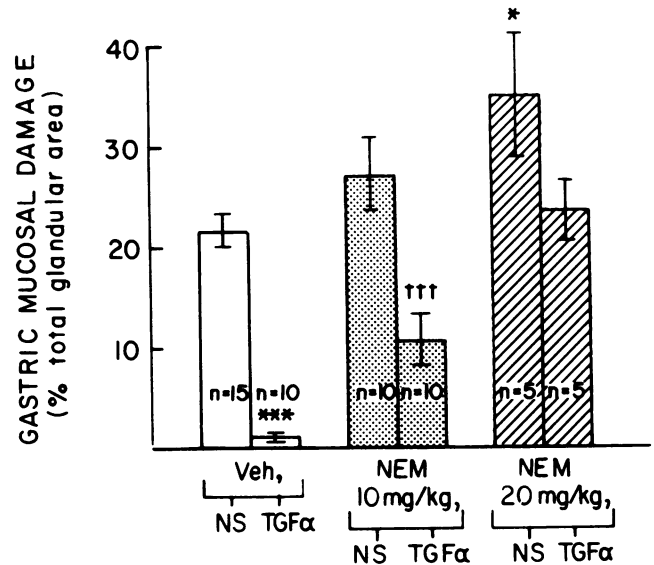


Figure 15. Effect of NEM on TGF α -induced protection against EtOH injury. Rats were pretreated with NEM (10 or 20 mg/kg) or distilled water (vehicle for NEM) subcutaneously; after 10 min, rats were given i.p. injection of TGF α (200 μ g/kg) or normal saline and then, after 30 min, both groups received orogastric 100% EtOH. Mean \pm SE of (n) rats per each study group. Veh, TGF α vs NEM 10 mg/kg, TGF α : *P* < 0.01; Veh, TGF α vs NEM 20 mg/kg, TGF α : *P* < 0.01; NEM 10 mg/kg, TGF α vs NEM 20 mg/kg, TGF α : *P* < 0.01. **P* < 0.05 vs Veh, NS; ****P* < 0.001 vs Veh, NS; †††*P* < 0.001 vs NEM 10 mg/kg, NS.

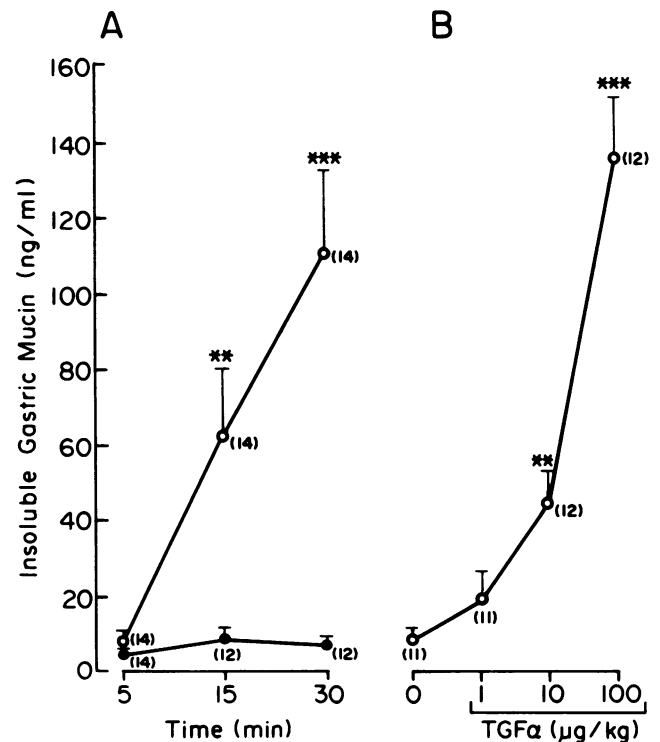


Figure 16. Effect of TGF α on gastric insoluble mucin. Insoluble gastric mucin was measured from lightly scraped gastric mucosa 5, 15, and 30 min after i.p. injection of TGF α (100 μ g/kg) or normal saline (control) (A) or 30 min after i.p. injection of TGF α (1–100 μ g/kg) or normal saline (control) (B). Mean \pm SE of (n) rats per each study group; ***P* < 0.01; ****P* < 0.001 vs normal saline control. ●, Normal saline; ○, TGF α 100 μ g/kg.

glutathione, or ODC-related events; (d) the protective effect of TGF α is temporally associated with activation of PLC- γ 1 and with a significant increase in adherent gastric mucin.

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References

1. Konturek, S. J., T. Radecki, T. Brzozowski, I. Piastucki, A. Dembinski, A. Dembinska-Kiec, A. Zumda, R. Gryglewski, and H. Gregory. 1981. Gastric cytoprotection by epidermal growth factor: role of endogenous prostaglandins and DNA synthesis. *Gastroenterology*. 81:438-443.
2. Konturek, S. J., T. Brzozowski, I. Piastucki, A. Dembinski, T. Radecki, A. Dembinska-Kiec, A. Zumda, and H. Gregory. 1981. Role of mucosal prostaglandins and DNA synthesis in gastric cytoprotection by luminal epidermal growth factor. *Gut*. 22:927-932.
3. Skov Olsen, P., S. S. Poulsen, P. Kirkegaard, and E. Nexø. 1984. Role of submandibular saliva and epidermal growth factor in gastric cytoprotection. *Gastroenterology*. 87:103-108.
4. Amagase, H., T. Murakami, M. Misaki, Y. Higashi, M. Ushijima, T. Fawa, and N. Yata. 1990. Protective effect of human epidermal growth factor against the experimental gastric mucosal lesions in rats. *Life Sci*. 47:1031-1036.
5. Marquardt, H., M. M. Hunkapiller, L. E. Hood, and G. J. Todaro. 1984. Rat transforming growth factor type I: structure and relation to epidermal growth factor. *Science (Wash. DC)*. 223:1079-1082.
6. Derynck, R., A. B. Roberts, M. E. Winkler, E. Y. Chen, and D. V. Goeddel. 1984. Human transforming growth factor- α : precursor structure and expression in *E. coli*. *Cell*. 38:287-297.
7. Todaro, G. J., C. Fryling, and J. E. DeLarco. 1980. Transforming growth factors produced by certain tumor cells: polypeptides that interact with epidermal growth factor receptor. *Proc. Natl. Acad. Sci. USA*. 77:5258-5262.
8. Massague, J. 1983. Epidermal growth factor-like transforming growth factor. II. Interaction with epidermal growth factor receptors in human placenta membranes and A431 cells. *J. Biol. Chem.* 258:13614-13620.
9. Carpenter, G., C. M. Stoscheck, Y. A. Preston, and J. E. DeLarco. 1983. Antibodies to the epidermal growth factor receptor block the biological activities of sarcoma growth factor. *Proc. Natl. Acad. Sci. USA*. 80:5627-5630.
10. Derynck, R. 1986. Transforming growth factor α : structure and biological activities. *J. Cell Biol.* 31:293-304.
11. Beauchamp, R. D., J. A. Barnard, C. M. McCutchen, J. A. Cherner, and R. J. Coffey, Jr. 1989. Localization of transforming growth factor α and its receptor in gastric mucosal cells. Implications for a regulatory role in acid secretion and mucosal renewal. *J. Clin. Invest.* 84:1017-1023.
12. Polk, W. H., P. J. Dempsey, W. E. Russell, P. I. Brown, R. D. Beauchamp, J. A. Barnard, and R. J. Coffey, Jr. 1992. Redistribution of transforming growth factor α in rat gastric mucosa following acute injury. *Gastroenterology*. 102:1467-1474.
13. Dworski, R., J. R. Sheller, N. E. Wickersham, J. A. Oates, K. L. Brigham, L. J. Roberts, and G. A. Fitzgerald. 1989. Allergen-stimulated release of mediators into sheep bronchoalveolar lavage fluid: effect of cyclooxygenase inhibition. *Am. Rev. Respir. Dis.* 139:46-51.
14. Whittle, B. J. R., G. A. Higgs, K. E. Eakins, S. Moncada, and J. R. Vane. 1980. Selective inhibition of prostaglandin production in inflammatory exudates and gastric mucosa. *Nature (Lond.)* 284:271-273.
15. Robert, A., F. W. Leung, D. G. Kaiser, and P. H. Guth. 1989. Potentiation of ASA-induced gastric lesions by exposure to cold in rats: role of acid secretion, mucosal blood flow, and gastric mucosal prostanoid content. *Gastroenterology*. 97:1147-1158.
16. Tietze, F. 1969. Enzymatic method for quantitative determination of nanogram amount of total and oxidized glutathione: applications to mammalian blood and other tissues. *Anal. Biochem.* 27:502-522.
17. Arteaga, C. L., M. D. Johnson, G. Todderud, R. J. Coffey, Jr., G. Carpenter, and D. L. Page. 1991. Elevated content of the tyrosine kinase substrate phospholipase C- γ 1 in primary human breast carcinomas. *Proc. Natl. Acad. Sci. USA*. 88:10435-10439.
18. Frackelton, A. R., Jr. 1985. Characterization of phosphotyrosyl proteins in cells transformed by Abelson murine leukemia virus: use of monoclonal antibody to phosphotyrosine. *Cancer Cells A Mon. Rev.* 3:339-345.
19. Wahl, M. I., T. O. Daniel, and G. Carpenter. 1988. Antiphosphotyrosine recovery of phospholipase C activity after EGF treatment of A-431 cells. *Science (Wash. DC)*. 241:968-970.
20. Bradford, M. A. 1979. A rapid and sensitive method for the quantitation of microgram quantities of protein utilizing the principle of dye binding. *Anal. Biochem.* 72:248-254.
21. Pegg, A. E., and S. McGill. 1979. Decarboxylation of ornithine and lysine in rat tissues. *Biochim. Biophys. Acta*. 568:416-427.
22. Boland, C. R., E. R. Kraus, J. M. Scheiman, C. Black, G. D. Deshmukh, and W. O. Dobbins III. 1990. Characterization of mucous cell synthetic functions in a new primary canine gastric mucous cell culture system. *Am. J. Physiol.* 258:G774-G787.
23. Duncan, D. B. 1955. Multiple range and multiple F tests. *Biometrics*. 11:1-42.
24. Robert, A., J. E. Nezamis, C. Lancaster, and A. J. Anchar. 1979. Cytoprotection by prostaglandins in rats: prevention of gastric necrosis produced by alcohol, HCl, NaOH, hypertonic NaCl, and thermal injury. *Gastroenterology*. 77:433-443.
25. Tarnawski, A., D. Hollander, H. Gergely, and J. Stachura. 1985. Comparison of antacid, cimetidine, and ranitidine in protection of the gastric mucosa against ethanol injury. *Am. J. Med.* 79(Suppl. 2C):19-23.
26. Davenport, H. W. 1967. Ethanol damage to canine oxyntic glandular mucosa. *Proc. Soc. Exp. Biol. Med.* 126:657-662.
27. Cooke, A. R. 1973. The role of acid in the pathogenesis of aspirin-induced gastrointestinal erosions and hemorrhage. *Am. J. Dig. Dis.* 18:225-237.
28. Wahl, M. I., S. Nishibe, S. Pann-Ghull, S. G. Rhee, and G. Carpenter. 1989. Epidermal growth factor stimulates tyrosine phosphorylation of phospholipase C-II independently of receptor internalization and extracellular calcium. *Proc. Natl. Acad. Sci. USA*. 86:1568-1572.
29. Nishibe, S., M. I. Wahl, S. G. Rhee, and G. Carpenter. 1989. Tyrosine phosphorylation of phospholipase C-II in vitro by the epidermal growth factor receptors. *J. Biol. Chem.* 264:10335-10338.
30. Konturek, P. K., T. Brzozowski, S. J. Konturek, and A. Dembinski. 1990. Role of epidermal growth factor, prostaglandin, and sulfhydryls in stress-induced gastric lesions. *Gastroenterology*. 99:1607-1615.
31. Szabo, S., J. S. Trier, and P. W. Frankel. 1981. Sulfhydryl compounds may mediate gastric cytoprotection. *Science (Wash. DC)*. 214:200-202.
32. Balint, G. A., and V. Varro. 1983. On the cytoprotective action of sulfhydryl-containing substances. *Acta. Physiol. Acad. Sci. Hung.* 60:139-142.
33. Strubelt, O., and R. Hoppenkamps. 1983. Relations between gastric glutathione and the ulcerogenic action of non-steroidal anti-inflammatory drugs. *Arch. Int. Pharmacodyn. Ther.* 262:268-278.
34. Deneke, S. M., and B. L. Fanburg. 1989. Regulation of cellular glutathione. *Am. J. Physiol.* 257:L163-L173.
35. Konturek, S. J., A. Dembinski, Z. Warzecha, W. Bielanski, T. Brzozowski, and D. Drozdowicz. 1988. Epidermal growth factor (EGF) in the gastroprotective and ulcer healing actions of colloidal bismuth subcitrate (De-Nol) in rats. *Gut*. 298:894-902.
36. Konturek, S. J., T. Brzozowski, W. Bielanski, Z. Warzecha, and D. Drozdowicz. 1989. Epidermal growth factor in the gastroprotective and ulcer-healing actions of sucralfate in rats. *Am. J. Med.* 86(Suppl 6A):32-37.
37. Tepperman, B. L., J. A. Kiernan, and B. D. Soper. 1989. The effect of sialoadenectomy on gastric mucosal integrity in the rat: roles of epidermal growth factor and prostaglandin E $_2$. *Can. J. Physiol. Pharmacol.* 67:1512-1519.
38. Elder, J. B., G. Williams, E. Lacey, and H. Gregory. 1978. Cellular localization of human urogastrone/epidermal growth factor. *Nature (Lond.)*. 271:466-467.
39. Heitz, P. U., M. Kasper, S. Van Noorden, J. M. Polak, H. Gregory, and A. G. E. Pearse. 1978. Immunohistochemical localisation of urogastrone to human duodenal and submandibular glands. *Gut*. 19:408-413.
40. Wright, N. A., C. Pike, and G. Elia. 1990. Induction of a novel epidermal growth factor-secreting cell lineage by mucosal ulceration in human gastrointestinal stem cells. *Nature (Lond.)*. 343:82-85.
41. Kasselberg, A. G., D. N. Orth, M. E. Gray, and M. T. Stahlman. 1985. Immunocytochemical localization of human epidermal growth factor/urogastrone in several human tissues. *J. Histochem. Cytochem.* 33:315-322.
42. Elder, J. B., C. Hiley, and H. Gregory. 1986. Epidermal growth factor-Urogastrone (hEGF-URO): localisation in the gut. *Can. J. Physiol. Pharm.* 5:148. (Abstr.).
43. Cartledge, S. A., and J. B. Elder. 1989. Transforming growth factor α and epidermal growth factor levels in normal human gastrointestinal mucosa. *Br. J. Cancer*. 60:657-660.
44. Derynck, R., A. B. Roberts, M. E. Winkler, E. Y. Chen, and D. V. Goeddel. 1984. Human transforming growth factor α : precursor structure and expression in *E. coli*. *Cell*. 38:287-297.

45. Chen, M. C., A. T. Lee, and A. H. Soll. 1991. Mitogenic response of canine fundic epithelial cells in short-term culture to transforming growth factor α and insulinlike growth factor I. *J. Clin. Invest.* 87:1716-1723.
46. Finke, U. M., M. Rutten, R. A. Murphy, and W. Silen. 1985. Effects of epidermal growth factor on acid secretion from guinea pig gastric mucosa: In vitro analysis. *Gastroenterology* 88:1175-1182.
47. Rhodes, J. A., J. P. Tam, U. Finke, M. Saunders, J. Bernanke, W. Silen, and R. A. Murphy. 1986. Transforming growth factor- α inhibits secretion of gastric acid. *Proc. Natl. Acad. Sci. USA* 83:3844-3846.
48. Lewis, J. J., J. A. Goldenring, I. M. Modlin, and R. J. Coffey. 1990. Inhibition of parietal cell H⁺ secretion by transforming growth factor alpha: a possible autocrine regulatory mechanism. *Surgery (St. Louis)* 108:220-227.
49. Konturek, S. J., M. Cieszkowski, J. Jaworek, J. Konturek, T. Brzozowski, and H. Gregory. 1984. Effects of epidermal growth factor on gastrointestinal secretions. *Am. J. Physiol.* 246:G580-G586.
50. Gregory, H., C. E. Thomas, J. A. Young, I. R. Willshire, and A. Garner. 1988. The contribution of the C-terminal undecapeptide sequence of urogastrone-epidermal growth factor to its biological action. *Regul. Pept.* 22:217-226.
51. Malden, L. T., U. Novak, and A. W. Burgess. 1989. Expression of transforming growth factor alpha messenger RNA in the normal and neoplastic gastrointestinal tract. *Int. J. Cancer* 43:380-384.
52. Scheving, L. A., R. A. Shiurba, T. D. Nguyen, and G. M. Gray. 1989. Epidermal growth factor receptor of the intestinal enterocyte. Localization to laterobasal but not brush border membrane. *J. Biol. Chem.* 264:1735-1741.
53. Mori, S., Y. Morishita, S. Sakai, S. Kurimoto, M. Okamoto, T. Kawamoto, and T. Kuroki. 1987. Electron microscopic evidence for epidermal growth factor receptor (EGF-R)-like immunoreactivity associated with the basolateral surface of gastric parietal cells. *Acta Pathol. JPN.* 37:1909-1917.
54. Thompson, J. 1988. Specific receptors for epidermal growth factor in rat intestinal microvillus membranes. *Am. J. Physiol.* 254:G429-G435.
55. Dembinski, A., H. Gregory, S. J. Konturek, and M. Polanski. 1982. Trophic action of epidermal growth factor on the pancreas and gastroduodenal mucosa in rats. *J. Physiol.* 325:35-42.
56. Slomiany, B. L., J. Liu, P. Yao, C. Y. Wu-Wang, J. P. Keogh, S. L. Wang, and A. Slomiany. 1990. Characterization of the epidermal growth factor receptor in the gastric mucosa. *Digestion* 47:181-190.
57. Konturek, S. J., T. Brzozowski, T. Radecki, I. Piastucki, and A. Dembinski. 1982. Cytoprotective effects of gastrointestinal hormones. In *Gut Peptides and Hormones*. A Miyoshi, editor. Biomedical Research Foundation, Tokyo. 411-417.
58. Schrieber, A. B., M. E. Winkler, and R. Derynck. 1986. Transforming growth factor α : a more potent angiogenic factor than epidermal growth factor. *Science (Wash. DC)* 232:1250-1253.
59. Gan, G. S., M. D. Hollenburg, K. L. MacCannel, K. Lederis, M. E. Winkler, and R. Derynck. 1987. Distinct vascular actions of epidermal growth factor-urogastrone and transforming growth factor- α . *J. Pharmacol. Exp. Ther.* 242:331-337.
60. Ibbotson, K. J., J. Harrod, M. Gowen, S. D'Souza, D. D. Smith, M. E. Winkler, R. Derynck, and G. R. Mundy. 1986. Human recombinant transforming growth factor α stimulates bone resorption and inhibits formation in vitro. *Proc. Natl. Acad. Sci. USA* 83:2228-2232.
61. Ebner, R., and R. Derynck. 1991. Epidermal growth factor and transforming growth factor- α : differential intracellular routing and processing of ligand-receptor complexes. *Cell Regul.* 2:599-612.
62. Carpenter, G., and M. I. Wahl. 1990. The epidermal growth factor family. In *Handbook of Experimental Pharmacology*. 95. Peptide Growth Factors and their Receptors I. M. B. Sporn and A. B. Roberts, editors. Springer Verlag, Berlin. 69-171.
63. Konda, Y., H. Nishisaki, O. Nakano, K. Matsuda, K. Wada, M. Nagao, T. Matozaki, and C. Sakamoto. 1990. Prostaglandin protects isolated guinea pig chief cells against ethanol injury via an increase in diacylglycerol. *J. Clin. Invest.* 86:1897-1903.
64. Berridge, M. J. 1984. Inositol trisphosphate and diacylglycerol as second messengers. *Biochem. J.* 220:345-360.
65. Silverstein, F. E., M. B. Kimmey, D. R. Saunders, C. M. Surawicz, R. A. Willson, and B. A. Silverman. 1987. Gastric protection by misoprostol against 1300 mg of aspirin. *Am. J. Med.* 83:32-36.
66. Griffin, M. R., W. A. Roy, and W. Shaffer. 1988. Non-steroidal anti-inflammatory drug use and death from peptic ulcer in elderly persons. *Ann. Intern. Med.* 109:359-363.
67. Tabor, C. W., and H. Tabor. 1984. Polyamines. *Ann. Rev. Biochem.* 53:749-790.
68. Mizui, T., and M. Doteuchi. 1983. Effect of polyamines on acidified ethanol-induced gastric lesions in rats. *JPN. J. Pharmacol.* 33:939-945.
69. Danzin, C., M. J. Jung, J. Grove, and P. Bey. 1979. Effect of α -difluoromethylornithine, an enzyme-activated irreversible inhibitor of ornithine decarboxylase, on polyamine levels in rat tissues. *Life Sci.* 24:519-524.
70. Romano, M., M. Razandi, A. Raza, S. Szabo, and K. J. Ivey. 1992. Cysteamine protects gastric epithelial cell monolayers against drug-induced damage: evidence for direct cellular protection by sulfhydryl compounds. *Gut* 33:30-38.
71. Boyd, S. C., H. A. Sasame, and M. R. Boyd. 1981. Gastric glutathione depletion and acute ulcerogenesis by diethylmaleate given subcutaneously to rats. *Life Sci.* 28:2987-2992.
72. Mutoh, H., H. Hiraishi, S. Ota, H. Yoshida, K. J. Ivey, A. Terano, and T. Sugimoto. 1990. Protective role of intracellular glutathione against ethanol-induced damage in cultured rat gastric mucosal cells. *Gastroenterology* 96:1452-1459.
73. Romano, M., M. Razandi, and K. J. Ivey. 1991. Role of sulfhydryl compounds in the defense of rat gastric epithelial cells against oxygen reactive metabolite-induced damage. *Ital. J. Gastroenterol.* 23:55-59.
74. Robert, A., D. Eberle, and N. Kaplowitz. 1984. Role of glutathione in gastric mucosal cytoprotection. *Am. J. Physiol.* 247:G296-G304.
75. Dupuy, D., and S. Szabo. 1986. Protection by metals against ethanol-induced gastric mucosal injury in the rat: comparative biochemical and pharmacologic studies implicate protein sulfhydryls. *Gastroenterology* 91:966-974.
76. Takeuchi, K., M. Okada, H. Niida, and S. Okabe. 1989. Role of sulfhydryls in mucosal injury caused by ethanol: relation to microvascular permeability, gastric motility and cytoprotection. *J. Pharmacol. Exp. Ther.* 248:826-841.
77. Morris, G. P., and P. L. Harding. 1984. Mechanisms of mucosal recovery from acute gastric damage: the roles of extracellular mucus and cell migration. In *Mechanisms of Mucosal Protection in the Upper Gastrointestinal Tract*. A. Alen, G. Flemstrom, A. Garner, W. Silen, and L. A. Turnberg, editors. Raven Press, Ltd., New York. 209-214.
78. McQueen, S., A. Allen, and A. Garner. 1984. Measurement of gastric and duodenal mucus gel thickness. In *Mechanisms of Mucosal Protection in the Upper Gastrointestinal Tract*. A. Alen, G. Flemstrom, A. Garner, W. Silen, and L. A. Turnberg, editors. Raven Press, Ltd., New York. 215-221.
79. Morris, G. P. 1985. The myth of the mucus barrier. *Gastroenterol. Clin. Biol.* 9:106-107.
80. Davenport, H. W. 1967. Salicylate damage to the gastric mucosal barrier. *N. Engl. J. Med.* 276:1307-1312.
81. Lacey, E. R. 1985. Gastric mucosal resistance to a repeated ethanol insult. *Scand. J. Gastroenterol.* 20(Suppl. 110):63-72.
82. Grisham, M. B., C. von Ritter, B. F. Smith, T. J. LaMont, and D. N. Granger. 1987. Interaction between oxygen radicals and gastric mucin. *Am. J. Physiol.* 253:G93-G96.
83. Pihan, G., C. Regillo, and S. Szabo. 1987. Free radicals and lipid peroxidation in ethanol- and aspirin-induced gastric mucosal injury. *Dig. Dis. Sci.* 32:1395-1401.